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TESTING RESIDUES: AN EXPERIMENTAL APPROACH

University of the Witwatersrand

TESTING RESIDUES: AN EXPERIMENTAL APPROACH

PhD thesis

In fulfilment of the requirements for the degree Doctor of Philosophy
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2009

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in Middelburg

DECLARATION

I declare that this thesis is my own unaided work. It is submitted for the degree of Doctor of Philosophy to the Faculty of Science at the University of the Witwatersrand, Johannesburg. It has not been submitted before for any other degree or examination at any other university.

Singned
Geeske Henriëtte Janine Langejans

day of 2009

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1 INTRODUCTION

In this thesis I investigate the preservation mechanisms for microscopic organic residues. In the first part of this Introduction I discuss the background of residue analysis and experimental archaeology and focus on current research issues regarding residue analysis. In the second part I discuss the research goal and sub-questions, which I answer in the course of this thesis. In last section I present the research design and discuss how I plan to answer the sub-questions. In this thesis the term residue analysis means " microscopic stone-tool residue analysis" (from Haslam 2006a). I also refer to the residue specialist as "he"; however this stands for "he or she".

1.1 BACKGROUND TO RESIDUE ANALYSIS AND EXPERIMENTAL ARCHAEOLOGY AND RELATED RESEARCH ISSUES

Residue analysis aims to identify microscopic remains or traces that are left on a tool's surface after use. Analysts have identified organic remains such as plant starch grains, fibres, tissue cells, resin; animal blood films and red blood cells, muscle tissue, connective fibres, bone fragments, hairs; fish scales and shell. Besides organic residues, it is also possible to distinguish inorganic deposits such as ochre and ash. Residue analysis is conducted with a light microscope, using a range of magnifications (between 50 and 800 times), a large experimental comparative collection and sometimes an ethno-archaeological comparative collection (Bruier 1976, Miller 1979, Anderson 1980, Fullagar, Furby and Hardy 1996, Loy 1997, Fullagar, Meehan and Jones 1999, Williamson 2000a, Williamson 2000b, Rots and Williamson 2004). Residue analysis, unlike micro-wear analysis, is not based on the edge damage created by use, but it involves direct evidence of portions of the processed materials, which adhere to the tools.

Residue analysis is a relatively young discipline. Although Von Stokar already discussed the preservation of organic residues on European prehistoric artefacts in 1938, it was after Zimmerman's (1973) discovery of red blood cells on a 2000 year

old mummy, that residue analysis began. Bruier (1976) and Shafer and Holloway (1979) were among the first to conduct a larger-scale residue analysis on stone tools. The early debate in the 1980s about residue analysis came from an interest in blood residues and, later, DNA analysis and the feasibility of these analyses (Anderson 1980, Mullis and Faloona 1987, Custer, Igenfritz and Doms 1988, Loy and Wood 1989, Loy and Hardy 1992, Smith and Wilson 1992, Eisele *et al.* 1995, Garling 1998, Haslam 2006a); (See Chapter Two for a detailed discussion of blood residue analysis). Nowadays, the identification of processed materials leads to a wider variety of other archaeological research interests. Fullagar (Fullagar 2006) and Haslam (Haslam 2006a) distinguish three levels of study for residue (and use-wear) analysis:

1. Determining the function of a particular tool or tool type and assessing viability of functional typologies (*e.g.* Dominguez- Rodrigo *et al.* 2001, Hurst *et al.* 2002, Lombard 2005, Hardy, Bolus and Conard 2008).
2. Assessing site function, mobility and the range of activities undertaken at particular places (*e.g.* Bruier 1976, Staller and Thompson 2002, Fullagar and Jones 2004, Piperno *et al.* 2004, Horrocks and Nunn 2007).
3. Evaluating theory and archaeological explanations of cultural change and stability (*e.g.* Fullagar and Field 1997, Connan and Nissenbaum 2003, Lombard 2008).

In addition, Haslam (Haslam 2006a) recognises two slightly more separate fields of interest where residue analysis:

4. Acts as a source of materials for further investigation, for example DNA analysis (*e.g.* Nelson *et al.* 1986, Hardy, Raff and Raman 1997, Williamson 2000b).
5. Investigates old and rare artefacts for further information (*e.g.* Loy 1998b, Loy and Dixon 1998, Barton 2007).

For example, residue analysis can help reconstruct hafting techniques of Middle Stone Age points and lead to new ideas regarding hunting strategies, the introduction and impact of new plant foods and the use of particular tool types (Staller and Thompson 2002, Lombard 2005, Hardy, Bolus and Conard 2008).

The results from residue studies are not always straightforward. Many studies use inadequately small samples to answer large questions (*e.g.* Barton and J.P. 1993,

Hardy 1999, Dominguez- Rodrigo *et al.* 2001, Hardy *et al.* 2001)[1] (but also see Lombard 2008). In particular, residue studies of small samples do not always have clear research aims. The studies are done out of a general interest and "to see if residues are present on tools" (*e.g.* Loy, Spriggs and Wickler 1992, Hardy 1999, Barton 2007). As a result these studies stand on their own and cannot be compared to other work so that their usefulness can be questioned. The lack of well-defined, large-scale research also means that there is no "significant body of results" (Haslam 2006a, 408). Without proper large-scale research this "body of results" is not formed and residue specialists can only accurately identify agency in small or single piece samples (*e.g.* Haslam 2006a, 412-415). This body of results can be used as a reference system, for example to deal with three remaining research issues. Grace (Grace 1996) and (Haslam 2006a) describe these three remaining issues (Figure 1.1):

1. Non-use related residues might contaminate artefacts.
2. Researchers are still unable to describe the mechanism of residue preservation.
3. Residues on a tool could be a palimpsest of events and it is impossible to distinguish these different events.

My study does not investigate the last point, but Haslam (2006a) discusses the use of agency as a possible means to overcome this issue. During deposition in sediments-, and during and after excavation, a tool can become contaminated with sediment components and modern contaminants such as ink and finger grease. In addition, unintentional events in prehistory may have led to non-use related residues adhering to a tool. Consequently, residue analysts could potentially misidentify contaminants for ancient use-related residues. Although some paint residues have been radiocarbon dated (Loy *et al.* 1990, Loy 1993), the age of residues on stone tools generally remains unknown[2]. In response to Grace (Grace 1996), Barton, Torrence and Fullagar. (1998) addressed the first point and demonstrate that starch grains from soils only contaminate tools in small quantities and that micro-wear analysis helps to discriminate use-related and contaminant residues. Similar experiments by

[1] Although Hardy *et al.* (2001) analyse 50 tools, the tools originate from two sites and cover eight typological groups. It is complicated to draw large-scale behavioural conclusions from such a diffuse sample.

[2] Dating of rock art is difficult because the stone matrix and the soot of recent fires interferes and contaminates the paint residues.

Williamson (2006) show that soil organics can adhere to stone tools, but usually do so in smaller quantities than use-related residue. In addition Wadley and Lombard (Lombard and Wadley 2007, Wadley and Lombard 2007) demonstrate that the association and distribution of residues are helpful in this matter. By compiling detailed maps of the residues on analysed tools they are able to rule out contaminants from their analysis. Use-related residues form a meaningful pattern on the tools, whereas contaminants tend to be randomly spread over the surface. Earlier, Loy, Spriggs and Wickler (1992) also observed that the distribution and association of residues must be meaningful in order to draw conclusions. This is a valuable approach, but herein also lays a problem, because it presupposes that the working edge is correctly identified (Grace 1996) and that enough experimental knowledge is gathered to predict meaningful residue patterns. A further investigation of distribution and association of residues could test whether Lombard and Wadley's approach is valuable without extensive knowledge of tool types and potentially used edges.

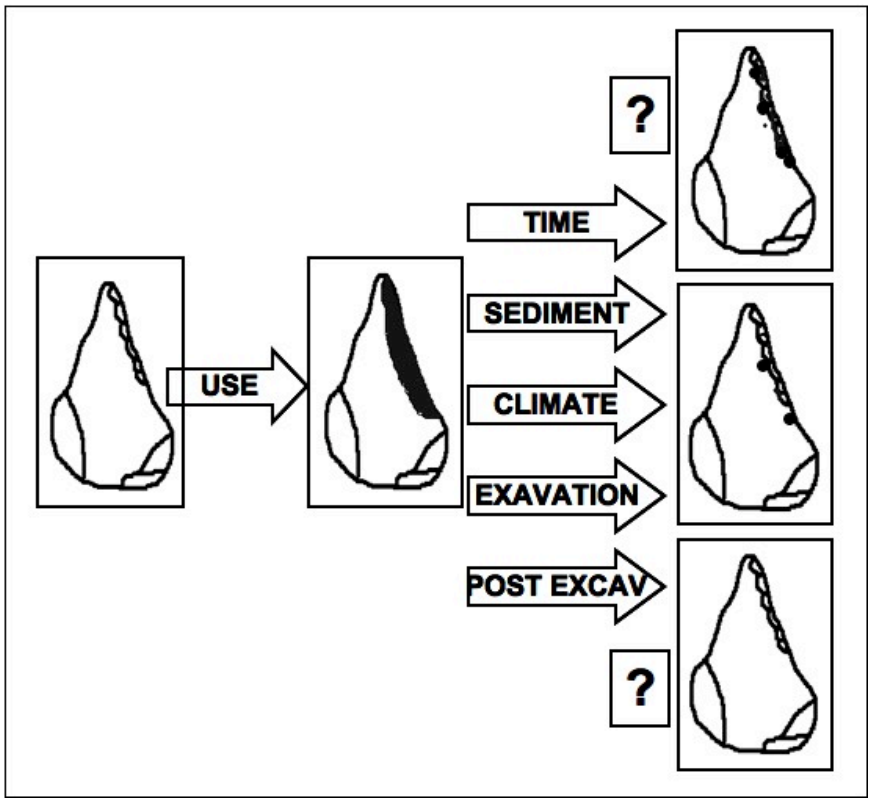


Figure 1.1: Schematic overview of some issues regarding residue preservation.

Regarding his concerns about residue preservation, Grace (1996, 214) observes that residue analysts "… simply record a phenomenon (…) and then make functional

interpretations by analogy without having explained the process that gave rise to the phenomenon. Odell (2001, 63) also argues that residue analysts "are inclined to consider what they see, not what is missing". This is slightly one sided as a few analysts observed discrepancies in their samples and noted differential preservation (*e.g.* Rots and Williamson 2004, Haslam, 2004 #128).

Some research into the preservation mechanisms of starch and blood has been conducted. Haslam (2004) and Barton and Matthews (2006) discuss the preservation mechanism for starch grains and predict that starch could preserve for long periods if particular stable circumstances are in place (see Chapter Three). Lu (2003, 2006) used experimental archaeology to show that unprotected starch does not preserve in sub-tropical environments. The combination of these studies demonstrates that starch only preserves under special circumstances. Research on blood shows that blood does not preserve well, but dry, desiccated conditions could allow some preservation (Gurfinkel and Franklin 1988, Cattaneo *et al.* 1991, Smith and Wilson 2001). Although there have been many investigations into molecular decay of other organic residues, thus far no research has been conducted on the preservation of other micro-remains (for an overview see Evershed 2008).

Barton *et al.* (Barton, Torrence and Fullagar 1998, 1231) consider that answers regarding the mechanisms of residue preservation "lie in the realm of biochemistry rather than archaeology" (also see Fullagar 1988). Although biochemistry can predict preservation and deterioration, it can say little about the state of preservation (*e.g.* Ambler and Daniel 1991, Eglinton and Logan 1991). Because these predictions give a residue specialist an idea of what could potentially preserve at a site, predictions regarding molecular preservation are valuable. Additionally, there are many archaeological studies that successfully employ bio-molecular analysis to identify molecular archaeological residues on pottery and stone tools (for an overview see Thomas 1993a, for more recent examples see Reuther *et al.* 2006, Mukherjee, Gibson and Evershed 2008). Note that molecular analysts struggle to answer archaeological questions (Barnard *et al.* 2007). Residue analysis depends on microscopy and has a strong visual aspect and although molecules could preserve, this does not mean that they are identifiable to a residue specialist. Through experimental archaeology and microscopy, analysts can learn the appearance of ancient residues and whether residues are preserved in a way that allows them to be recognised. Experimental archaeology can be a tool for devising middle-range theories (Binford 1983, Raab and

Goodyear 1984, Schiffer 1988). It permits residue specialists to bridge the gap between the apparently static archaeological data to a dynamic reconstruction of past processed materials. In doing so, experimental studies give residue analysis a framework, which allows predictions to be made about residue preservation, recognition and possible misinterpretation.

Because archaeologists need to be aware of potential biases in results, it is necessary to understand the mechanism of preservation. Wadley, Lombard and Williamson (2004), for example, suggest that plant type residues preserve better than animal type residues and that in an acidic environment blood residues might decay faster than plant residues. In order to avoid misinterpretation of processed materials and residue distribution patterns one needs to understand whether certain residues might be missing in an analysis because of uneven preservation. This leads to two general questions:

1. Do organic residues preserve?
2. Do particular residues preserve better than others and if so under which circumstances?

Understanding of the general preservation mechanism, under points 1 and 2, allows for preservation predictions at site level. Predictions should answer two main questions:

3. What types of sites allow (partial) residue preservation?
4. What residue types could be missing from a particular site?

1.2 RESEARCH DESIGN

This study includes a large-scale experiment to investigate the mechanisms of residue preservation and contamination issues. I used replicated stone tools and residues and compared them to ancient tools from the South African Earlier Stone Age and Middle Stone Age. With this large-scale research I wish to contribute to the “significant body of results”, referred to earlier, which is needed in order to use residue analysis to answer large questions of resource exploitation and changing social and ecological involvement (Haslam 2006a).

I used experimental archaeology in an attempt to answer the questions posed in the previous paragraph. Experimentation is part of a "hypothetico-deductive" process. If a hypothesis resists falsification and is support by experimentation, then it can be regarded valid (Outram 2008, 1). In this light, experimental archaeology is a scientific research method and as such it should answer clear research questions, uphold rigorous standards and be embedded in an academic context (Outram 2008). Experimental (also called actualistic) archaeology comprises five major classes of experiments (Reynolds 2006, Outram 2008):

1. Construct
2. Processes and function experiments
3. Simulation
4. Eventuality trial
5. Technological innovation

This study falls into class number 3. Simulation experiments generally investigate formation processes of the archaeological record and post-depositional processes (O'Connell *et al.* 2001, Reynolds 2006).

From the four general questions in the previous paragraph, I formulated eight sub-questions about contamination and preservation mechanisms, which I aim to answer in this thesis.

Questions concerning contamination issues:

1. What is the role of contamination by extraneous residues occurring in the archaeological sediments and as the result of post-excavation handling? Is there such contamination?
2. Do residues move from the sediment onto the stone tools?
3. Can contaminants be distinguished from use-related residues?
4. Is the distribution and association approach by Lombard and Wadley helpful to distinguish use-related residues from contaminants?

Questions concerning the process of decay:

5. Do muscle tissue, bone, fat, blood and plant starch grains and plant tissue preserve in the same manner?

6. What is the influence of time, precipitation, temperature, burial circumstances, soil pH, soil constitution, rock type, and processed material on the preservation of residues?
7. Can I make predictions about site suitability for analysis and preservation biases from points 5 and 6?
8. Are residues representative of past processed materials?

In order the answer these questions, I designed the following project: I made experimental stone tools from chert, hornfels and flint and used them on animal and plant products to create muscle tissue, bone, fat, plant starch grains and plant tissue residues. I also applied fat, blood and starch grains to sandpaper pieces with varying grain size. The next step was to deposit the samples at four locations, Sterkfontein and Sibudu in South Africa, and Zelhem and Wilhelmina Polder in the Netherlands. The deposition varied from buried, about 5 centimetres under the surface, to exposed, lying on top of the soil surface. At the two South African cave sites, some of the tools were deposited inside the cave and others outside the cave. One part of the South African samples was left to weather for four weeks and the other part for one year. The Dutch samples were left to weather for six months. After these intervals, I excavated the samples and analysed them with a microscope for residues. Archaeological tools from Sterkfontein and Sibudu were also analysed for residues. I hoped to find a correlation between these ancient residues and the residues from the experimental sample.

In Chapter Two I discuss the experiments, sites and methodology in detail. In addition to the practical analysis of (experimental) residues, this thesis also includes a literature review of the research on preservation of archaeological remains. It summarises general degradation mechanisms and discusses the optimal preservation circumstances for plant tissue, starch, resin, blood, bone, fat and hair. Chapter Three sets this study in the wider context of preservation studies and reviews the literature. Chapter Three also describes the archaeological, experimental and environmental settings in details. This leads to predictions about what residue types are expected to preserve at which locality. The results from the residue analyses are presented in Chapters Four to Six. In Chapter Seven I discuss the results and answer the sub-questions posed above.

2 METHODOLOGY

This chapter starts with a discussion of the experimental and archaeological samples and sites. This is followed by a presentation of the methodology, including microscopy, the use of the contextual approach and the preservation index, used to quantify the preservation of residues. Subsequently, I discuss the identification criteria for residues.

2.1 THE SAMPLES

Large-scale experiments were conducted to create tools with residues and to mimic post-depositional weathering and residue decay. The experimental tools were analysed with a microscope for residues. Archaeological samples were also analysed for residues. The results from the experiments and the analysis of the archaeological tools are intended to answer the research questions posed in Chapter One. Experiments were conducted at Sibudu Cave and Sterkfontein Caves, South Africa, and at Wilhelmina Polder and Zelhem, in The Netherlands. The first two locations are archaeological sites and samples of archaeological tools from these sites were also analysed. I discuss the environment and sediment at these locations and predict how this influences residue preservation.

The depositional circumstances differ greatly between the two archaeological assemblages from Sterkfontein and Sibudu, which is why these sites were selected. The artefacts from Sibudu, with an age of about 70 and 60 ka old, are much younger than those from Sterkfontein (about 1,5 Ma) and the material at Sibudu was deposited inside the shelter and was therefore protected rather than exposed, as was the case at Sterkfontein. The experiments at Zelhem and Wilhelmina Polder are open-air sites with high annual precipitation and this contrasts the South African sites.

2.1.1 Sterkfontein sample

The Sterkfontein Caves are situated about 60 kilometres north of Johannesburg, in Gauteng province, South Africa (see Figure 2.1). The site is located in the Blaaubank

River Valley and the modern Blaaubank River is about 500 metres away (Kuman and Clarke 2000, Avery 2001). Sterkfontein is part of South Africa's Cradle of Humankind UNESCO world heritage site. Other sites in the area are also part of the Cradle of Humankind and they are comparable in age and geological setting.

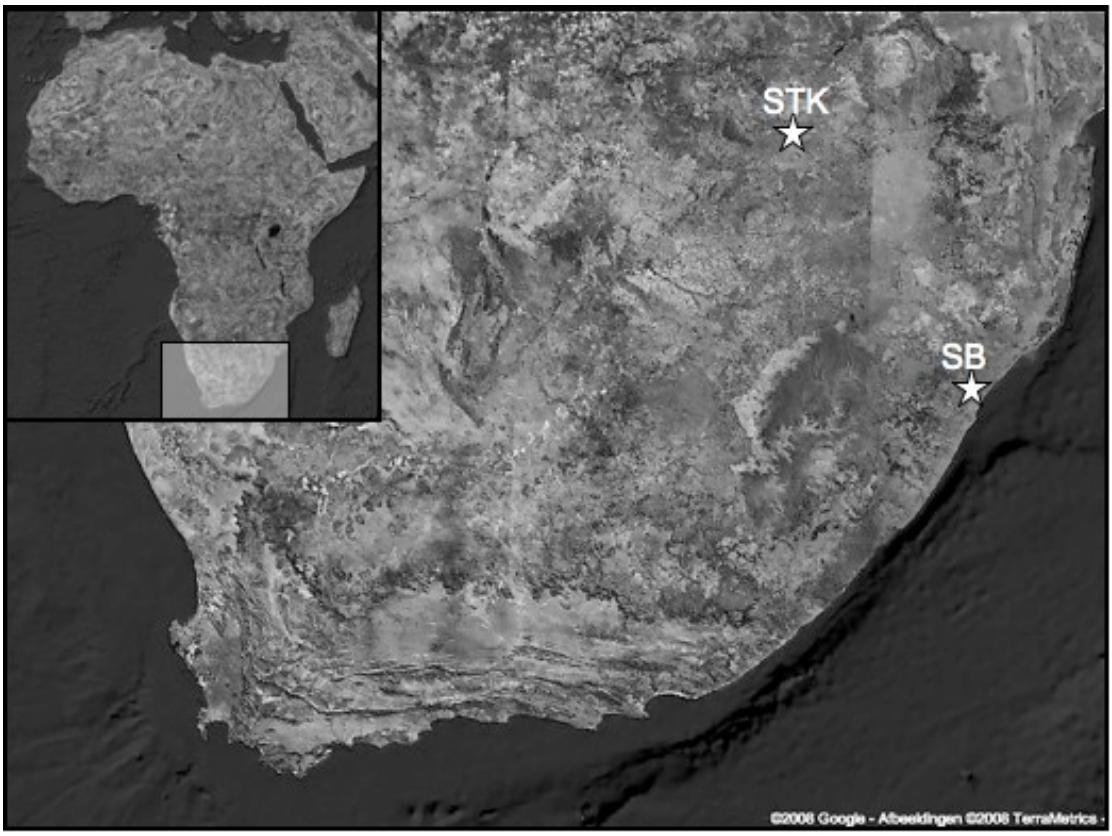


Figure 2.1: Map with the locations of Sibudu Cave and the Sterkfontein Caves. STK= Sterkfontein; SB= Sibudu (www.googlemaps.com 2008).

The underground cave systems in the Cradle are karst caverns in dolomite. In an ongoing process groundwater leaches out parts of the weaker karst creating caverns (Partridge 2000). Later, the slightly acidic groundwater erodes narrow shafts, which openes the caves to surface. From then on, material including sediment and biological remains can wash into the caves. Once in the cave, the material in the debris mound, including rock spalls, under the entrance calcifies and formes a cemented matrix popular known as breccia. In some archaeological stratigraphic levels, groundwater has decalcified breccia, creating a soft matrix (Kuman and Clarke 2000, Partridge 2000). Cave formation is discussed in detail in section 3.3.1. Because it is deposited from the surface into these underground caves, the archaeological material from Sterkfontein is not in primary context.

The Sterkfontein Caves capture a periods of infill from about ~3,5 Ma to ~113,500 years ago (Partridge 2000, Reynolds, Clarke and Kuman 2007). The sedimentary breccia deposits are divided into Members (M), ranging from the oldest, M1, to the youngest, M6, followed by a Post-Member 6 Infill. M5 and Post-Member 6 are the only deposits, which yielded tools; the other members only contained fossilised bone, although wood is preserved in M4. Thirty artefacts from M5 were analysed for this study. They are dated to about 1.5 Ma old and fall in the Early Acheulean industrial complex. In Chapter Three I discuss the present day and ancient environment and climate, which have an influence on residue preservation at Sterkfontein.

At Sterkfontein eight experimental samples, totalling 80 chert tools with residues, were deposited at four locations. Two locations are inside the cave, on a modern talus cone (see locations 1 and 2 in Figure 2.2), and two are outside the cave at the surface (see locations 3 and 4 in Figure 2.2). The location inside held both exposed tools, which were left on top the sediment and tools buried under about 5 centimetres of local sediment. One location outside the cave also held exposed tools and the other a buried sample. After four weeks, half of the tools were collected from the four locations. The remaining tools were collected after one year. This part of the experiment tests the influence of immediate burial on residue preservation, opposed to prolonged exposure, comparing potential differences in preservation at different locations within the site and the rate of residue deterioration over time.

The experimental tools were knapped specifically for this project and carefully handled to avoid contamination. Chert was used for the tools because part of the archaeological tools was also made on chert. In addition, chert is granular and comparable to hornfels used in the Sibudu experiments. It was therefore selected over quartz although this stone was also used by hominids to make tools. Before deposition, the tools in this study were used to work four organic materials to create experimental residues. These materials include: potato, to create starch residues; fresh wood, to create woody tissue residues; bone, to create fatty bone and bony residues; and muscle tissue, to create muscle tissue residues. In addition, a sample of unused flakes was included. This part of the experiment tests the preservation of different residue types in the situations described above. Additionally, the experiment tests whether these use-related residues leave a meaningful distribution pattern on the tool surface. It also tests what sediment organics, as contaminants, adhere to tools and what pattern these

residues leave on the tool surface. Table 2.1 presents an overview of the Sterkfontein experiments. The experimental tools from Sterkfontein were carefully collected by myself and stored in plastic ziplock bags. They were not washed prior to analysis and only handled by myself, avoiding post-excavation contamination.

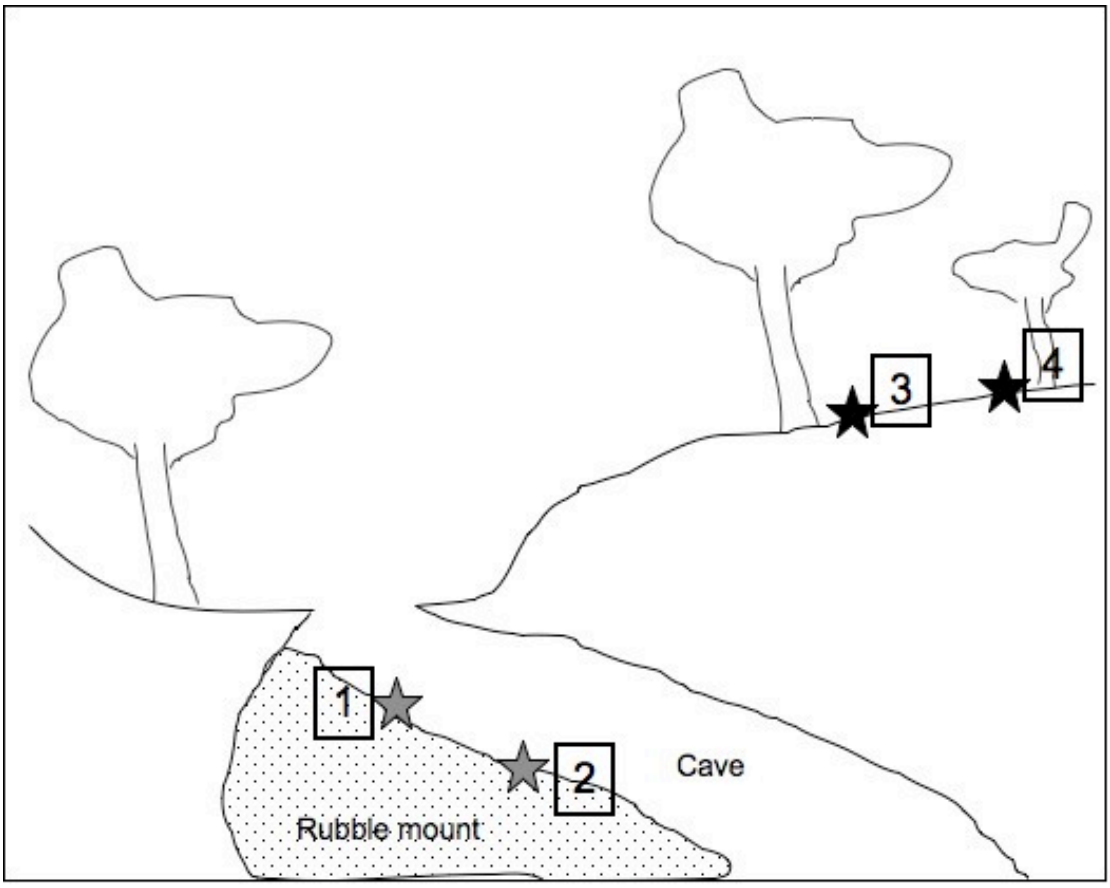


Figure 2.2: Schematic overview of the four locations of the experiments and measuring stations at Sterkfontein. The grey stars are the locations inside the cave and the black stars are situated outside the cave. 1= soil moisture station up the rubble slope; 2= soil moisture station down the rubble slope; 3= soil moisture station outside the cave, down hill; 4= = soil moisture station outside the cave, up hill.

Table 2.1: Overview of the Sterkfontein experimental sample.

Tool number	Residue type	Raw material;	Location	Situation	Period
STK81	Starch	Chert	Inside	Exposed	One year
STK82	Starch	Chert	Inside	Exposed	One year
STK83	Starch	Chert	Inside	Exposed	Four weeks
STK84	Starch	Chert	Inside	Exposed	Four weeks
STK85	Starch	Chert	Inside	Buried	One year
STK86	Starch	Chert	Inside	Buried	One year
STK87	Starch	Chert	Inside	Buried	Four weeks
STK88	Starch	Chert	Inside	Buried	Four weeks
STK89	Starch	Chert	Outside	Exposed	One year
STK90	Starch	Chert	Outside	Exposed	One year
STK91	Starch	Chert	Outside	Exposed	Four weeks
STK92	Starch	Chert	Outside	Exposed	Four weeks
STK93	Starch	Chert	Outside	Buried	One year
STK94	Starch	Chert	Outside	Buried	One year
STK95	Starch	Chert	Outside	Buried	Four weeks
STK96	Starch	Chert	Outside	Buried	Four weeks
STK97	Wood	Chert	Inside	Exposed	One year
STK98	Wood	Chert	Inside	Exposed	One year
STK99	Wood	Chert	Inside	Exposed	Four weeks
STK100	Wood	Chert	Inside	Exposed	Four weeks
STK101	Wood	Chert	Inside	Buried	One year
STK102	Wood	Chert	Inside	Buried	One year
STK103	Wood	Chert	Inside	Buried	Four weeks
STK104	Wood	Chert	Inside	Buried	Four weeks
STK105	Wood	Chert	Outside	Exposed	One year
STK106	Wood	Chert	Outside	Exposed	One year
STK107	Wood	Chert	Outside	Exposed	Four weeks
STK108	Wood	Chert	Outside	Exposed	Four weeks
STK109	Wood	Chert	Outside	Buried	One year
STK110	Wood	Chert	Outside	Buried	One year
STK111	Wood	Chert	Outside	Buried	Four weeks
STK112	Wood	Chert	Outside	Buried	Four weeks
STK113	Animal	Chert	Inside	Exposed	One year
STK114	Animal	Chert	Inside	Exposed	One year
STK115	Animal	Chert	Inside	Exposed	Four weeks
STK116	Animal	Chert	Inside	Exposed	Four weeks
STK117	Animal	Chert	Inside	Buried	One year
STK118	Animal	Chert	Inside	Buried	One year
STK119	Animal	Chert	Inside	Buried	Four weeks
STK120	Animal	Chert	Inside	Buried	Four weeks
STK121	Animal	Chert	Outside	Exposed	One year
STK122	Animal	Chert	Outside	Exposed	One year
STK123	Animal	Chert	Outside	Exposed	Four weeks
STK124	Animal	Chert	Outside	Exposed	Four weeks
STK125	Animal	Chert	Outside	Buried	One year
STK126	Animal	Chert	Outside	Buried	One year
STK127	Animal	Chert	Outside	Buried	Four weeks
STK128	Animal	Chert	Outside	Buried	Four weeks
STK129	Bone	Chert	Inside	Exposed	One year
STK130	Bone	Chert	Inside	Exposed	One year
STK131	Bone	Chert	Inside	Exposed	Four weeks
STK132	Bone	Chert	Inside	Exposed	Four weeks
STK133	Bone	Chert	Inside	Buried	One year

Continuation Table 2.1: Overview of the Sterkfontein experimental sample.

Tool number	Residue type	Raw material;	Location	Situation	Period
STK134	Bone	Chert	Inside	Buried	One year
STK135	Bone	Chert	Inside	Buried	Four weeks
STK136	Bone	Chert	Inside	Buried	Four weeks
STK137	Bone	Chert	Outside	Exposed	One year
STK138	Bone	Chert	Outside	Exposed	One year
STK139	Bone	Chert	Outside	Exposed	Four weeks
STK140	Bone	Chert	Outside	Exposed	Four weeks
STK141	Bone	Chert	Outside	Buried	One year
STK142	Bone	Chert	Outside	Buried	One year
STK143	Bone	Chert	Outside	Buried	Four weeks
STK144	Bone	Chert	Outside	Buried	Four weeks
STK145	Blank	Chert	Inside	Exposed	One year
STK146	Blank	Chert	Inside	Exposed	One year
STK147	Blank	Chert	Inside	Exposed	Four weeks
STK148	Blank	Chert	Inside	Exposed	Four weeks
STK149	Blank	Chert	Inside	Buried	One year
STK150	Blank	Chert	Inside	Buried	One year
STK151	Blank	Chert	Inside	Buried	Four weeks
STK152	Blank	Chert	Inside	Buried	Four weeks
STK153	Blank	Chert	Outside	Exposed	One year
STK154	Blank	Chert	Outside	Exposed	One year
STK155	Blank	Chert	Outside	Exposed	Four weeks
STK156	Blank	Chert	Outside	Exposed	Four weeks
STK157	Blank	Chert	Outside	Buried	One year
STK158	Blank	Chert	Outside	Buried	One year
STK159	Blank	Chert	Outside	Buried	Four weeks
STK160	blank	Chert	Outside	Buried	Four weeks

Loy (1998a) and Willamson (2000b) conducted residue analysis on artefacts from M5. They found organic residues, including starch, plant fibres and blood on tools, making them the oldest residues in the world. In the archaeological sample I analyse, nine artefacts overlap with their work (see Table 2.2). I analysed 30 artefacts, made on a variety of stone materials, including quartz and chert. The tools are a random selection from the Early Acheulean assemblage and include cores and flakes. My selection is comparable to Loy's selection. This part of the study tests if there is a correlation in the preservation of ancient residues and the experimental residues.

The curation history of the Sterkfontein archaeological sample is complex. Many tools were extracted from the sieve during wet sieving, which is standard practice. Once excavated/recovered from the soft breccia deposits, the tools were washed and stored on-site in cardboard boxes. The tools were then transported to the laboratory at the University of the Witwatersrand where they underwent cleaning, numbering and analysis. During the stone tool analysis, no gloves are used. Some cores and all core tools from Sterkfontein were moulded and cast (Clarke pers. comm.

2007), including the biface, cleaver, and core in my sample. For this process the tool is first dusted with talcum powder and then a layer of silicon rubber is applied. During analysis and casting, which took some years, the tools sometimes sat in the open laboratory at Wits or in drawers and stacked wooden trays. Note that the flourmill in Braamfontein is not far from the laboratory and that the windows are often open. After analysis and casting, the Sterkfontein tools were stored in drawers on top of textiles. Precious pieces were sometimes wrapped in velvet textiles. The tools were often handled and some of them drawn, and no preventive measures were taken to avoid contamination (Kuman pers. comm. 2007 and personal observation).

Loy and Williamson analysed the tools after they were placed in storage in the drawers. The casting rubber is greasy and fatty. It is most likely that both the talcum powder and rubber left micro-remains on the tool surface in the form of starch grains and grease/fat deposits respectively. Starch grains from the nearby flour mill probably contaminated the sample. Skin scales or flakes, which under the microscope look similar to bone-like flakes or deposits, also appear on handled tools. Furthermore, handling leaves finger grease and modern plant fibres on the tools and contamination from successive handling, is also not unlikely. This is how, for example, starch grains can move from tool to tool. Although it is unwise to analyse a sample, which lacks integrity, I included this Sterkfontein sample because it is often used as an example of residues preserving for millions of years.

Table 2.2: Overview of the archaeological sample from Sterkfontein and the interpretation by Loy.

Tool number	Tool type	Deposit	Raw material	Cast	Loy's residues and interpretation
STK403	Flake	M5E-A	Quartz	-	-
STK2053	Biface	M5E-A	Chert	Yes	-
STK2504	Cortical flake	M5E-A	Quartz	-	Red blood cells and cellulose.
STK2630	Incomplete flake	M5E-A	Quartzite	-	-
STK2739	Flake	M5E-A	Quartzite	-	-
STK2808	Incomplete flake	M5E-A	Quartz	-	-
STK2982	Incomplete flake	M5E-A	Quartz	-	-
STK3049	Flake	M5E-A	Chert	-	-
STK3051	Flake	M5E-A	Quartzite	-	Bone and hair
STK3067	Incomplete flake	M5E-A	Quartz	-	-
STK3109	Flake	M5E-A	Quartz	-	-
STK3238	Retouched flake	M5E-A	Chert	-	-
STK3312	Flake	M5E-A	Quartz	-	Blood with red blood cells visible and bone.
STK3363	Flake	M5E-A	Quartz	-	-
STK3411	Flake	M5E-A	Quartzite	-	-
STK3977	Core	M5E-A	Quartz	Yes	Blood. Successful ancient DNA sequencing, unknown donor.
STK4883	Flake	M5E-A	Quartz	-	-
STK5872	Flake	M5E-A	Quartz	-	Bone and fat indicate use as a spokeshave or scraper on dry fresh bone.
STK5885	Flake	M5E-A	Quartz	-	Contaminants.
STK6048	Flake	M5E-A	Quartz	-	-
STK6049	Flake	M5E-A	Quartz	-	-
STK6085	Flake	M5E-A	Chert	-	Bone and connective fibres indicate bone working.
STK6121	Flake	M5E-A	Quartz	-	No residues
STK6129	Flake	M5E-A	Quartz	-	?
STK6230	Flake	M5E-A	Chert	-	?
STK7318	Flake	M5E-A	Chert	-	-
STK7975	Flake	M5E-A	Chert	-	-
STK8075	Flake	M5E-A	Quartz	-	-
STK8849	Flake	M5E-A	Quartzite	-	-
STK8855	Flake	M5E-A	Quartz	-	-
STK8903	Retouched flake	M5E-A	Quartz	?	-
STK9521	Flake	M5E-A	Chert	-	-
STK10363	Cleaver	M5E-A	Quartzite	Yes	Blood. Successful ancient DNA sequencing, hominid donor.

2.1.2 The Sibudu sample

Sibudu Cave is located about 40 kilometres north of Durban, KwaZulu-Natal province, South Africa (see Figure 2.1). The cave, which is more a shelter, is 15 kilometres from the Indian Ocean and situated on a steep cliff overlooking the Tongati River (Wadley and Jacobs 2006). Sibudu is located about 100 metres above mean sea-level.

The shelter has been worn out by fluvial action from the prehistoric Tongati River in a Natal Group sandstone cliff. The site is in primary context (Pickering 2006). The ashy sediments in the shelter are anthropogenic and the stratigraphy is reconstructed with centimetre to millimetre precision. At Sibudu bone remains and stone tools are found in abundance and the stratigraphic layers in which they are found are dated from the final Middle Stone Age (MSA) to a pre-Still Bay age (~37 ka to older than 75 ka) (Wadley and Jacobs 2006) and fall within Oxygen Isotope Stage (OIS) 4 and 3. Thus far no macro plant remains have been found. The archaeological tools used in this study are from the Howiesons Poort layers and are dated to >60 ka (Wadley and Jacobs 2006). In Chapter Three I discuss the present day and ancient environment and climate at Sibudu, which have an influence on residue preservation.

At Sibudu eight experimental samples, totalling 80 hornfels tools with residues, were deposited at four locations. Two locations are inside the shelter (see locations 3 and 4 in Figure 2.3) and two are at the dripline (see locations 1 and 2 in Figure 2.3). Of the two locations inside one held exposed tools, which were left on top of the sediment and at the other location experimental tools were buried under about 5 centimetres of local sediment. One location at the dripline also held exposed tools and the other a buried sample. After four weeks, half of the tools were collected from the four locations. The remaining tools were collected after one year. This part of the experiment tests the influence of immediate burial on residue preservation, opposed to prolonged exposure. It also tests potential differences in preservation at different locations within the site and the rate of residue deterioration over time.

The experimental tools were knapped specifically for this project and carefully handled to avoid contamination. Hornfels was used for the tools because part of the archaeological sample was also made on hornfels. In addition, hornfels is granular and comparable to chert used in the Sterkfontein experiments. Before deposition, the tools in this study were used to work four organic materials to create experimental residues. These materials included: potato, to create starch residues, fresh wood, to create woody tissue residues, bone, to create fatty bone and bony residues, and muscle tissue, to create muscle tissue residues. In addition, a sample of unused flakes was included. This part of the experiments tests the preservation of different residues in the situations described above. Additionally, it tests if these use-related residues leave a meaningful distribution pattern on the tool surface. It also tests what sediment organics, that is, post-depositional contaminants, adhere to tools and what pattern

these residues leave on the tool surface. Table 2.3 presents an overview of the Sibudu experiments. The experimental tools from Sibudu were carefully collected by myself and stored in plastic ziplock bags. They were not washed prior to analysis and only handled by myself, avoiding post-excavational contamination.

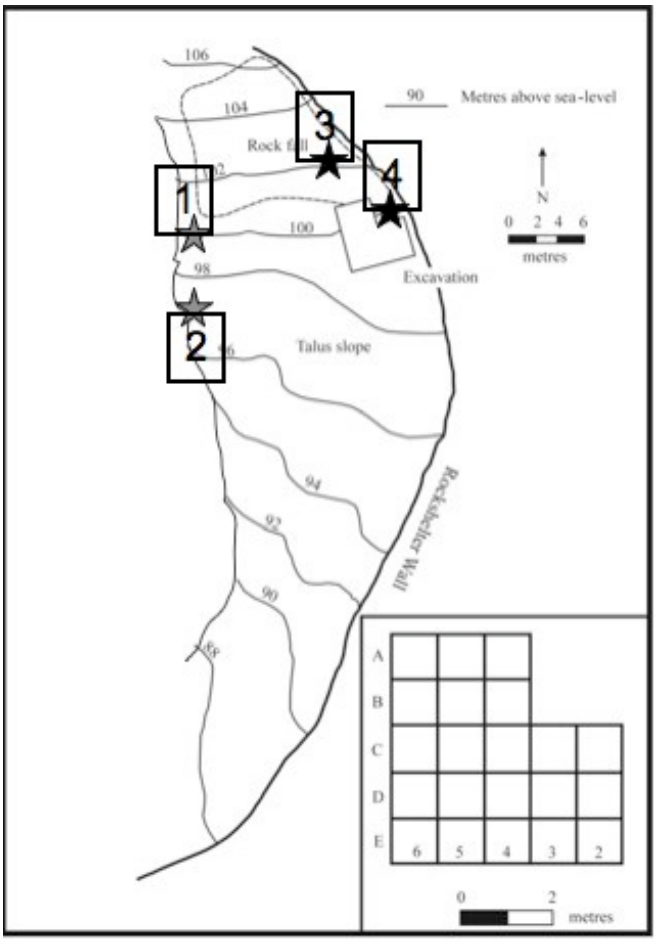


Figure 2.3: Map with four locations of the experiments and measuring stations at Sibudu Cave. The grey stars indicate the location of the dripline experiments; the black stars indicate the experiment locations inside the shelter. 1= soil moisture station at the dripline; 2= control soil moisture station at the dripline; 3= soil moisture station inside the shelter; 4= control soil moisture station inside the shelter.

Table 2.3: Table with an overview of the Sibudu experiments.

Tool number	Residue type	Raw material	Location	Situation	Period
SB81	Starch	Hornfels	Inside	Exposed	One year
SB82	Starch	Hornfels	Inside	Exposed	One year
SB83	Starch	Hornfels	Inside	Exposed	Four weeks
SB84	Starch	Hornfels	Inside	Exposed	Four weeks
SB85	Starch	Hornfels	Inside	Buried	One year
SB86	Starch	Hornfels	Inside	Buried	One year
SB87	Starch	Hornfels	Inside	Buried	Four weeks
SB88	Starch	Hornfels	Inside	Buried	Four weeks
SB89	Starch	Hornfels	Outside	Exposed	One year
SB90	Starch	Hornfels	Outside	Exposed	One year
SB91	Starch	Hornfels	Outside	Exposed	Four weeks
SB92	Starch	Hornfels	Outside	Exposed	Four weeks
SB93	Starch	Hornfels	Outside	Buried	One year
SB94	Starch	Hornfels	Outside	Buried	One year
SB95	Starch	Hornfels	Outside	Buried	Four weeks
SB96	Starch	Hornfels	Outside	Buried	Four weeks
SB97	Wood	Hornfels	Inside	Exposed	One year
SB98	Wood	Hornfels	Inside	Exposed	One year
SB99	Wood	Hornfels	Inside	Exposed	Four weeks
SB100	Wood	Hornfels	Inside	Exposed	Four weeks
SB101	Wood	Hornfels	Inside	Buried	One year
SB102	Wood	Hornfels	Inside	Buried	One year
SB103	Wood	Hornfels	Inside	Buried	Four weeks
SB104	Wood	Hornfels	Inside	Buried	Four weeks
SB105	Wood	Hornfels	Outside	Exposed	One year
SB106	Wood	Hornfels	Outside	Exposed	One year
SB107	Wood	Hornfels	Outside	Exposed	Four weeks
SB108	Wood	Hornfels	Outside	Exposed	Four weeks
SB109	Wood	Hornfels	Outside	Buried	One year
SB110	Wood	Hornfels	Outside	Buried	One year
SB111	Wood	Hornfels	Outside	Buried	Four weeks
SB112	Wood	Hornfels	Outside	Buried	Four weeks
SB113	Animal	Hornfels	Inside	Exposed	One year
SB114	Animal	Hornfels	Inside	Exposed	One year
SB115	Animal	Hornfels	Inside	Exposed	Four weeks
SB116	Animal	Hornfels	Inside	Exposed	Four weeks
SB117	Animal	Hornfels	Inside	Buried	One year
SB118	Animal	Hornfels	Inside	Buried	One year
SB119	Animal	Hornfels	Inside	Buried	Four weeks
SB120	Animal	Hornfels	Inside	Buried	Four weeks
SB121	Animal	Hornfels	Outside	Exposed	One year
SB122	Animal	Hornfels	Outside	Exposed	One year
SB123	Animal	Hornfels	Outside	Exposed	Four weeks
SB124	Animal	Hornfels	Outside	Exposed	Four weeks
SB125	Animal	Hornfels	Outside	Buried	One year
SB126	Animal	Hornfels	Outside	Buried	One year
SB127	Animal	Hornfels	Outside	Buried	Four weeks
SB128	Animal	Hornfels	Outside	Buried	Four weeks
SB129	Bone	Hornfels	Inside	Exposed	One year
SB130	Bone	Hornfels	Inside	Exposed	One year
SB131	Bone	Hornfels	Inside	Exposed	Four weeks
SB132	Bone	Hornfels	Inside	Exposed	Four weeks
SB133	Bone	Hornfels	Inside	Buried	One year

Continuation Table 2.3: Table with an overview of the Sibudu experiments.

Tool number	Residue type	Raw material	Location	Situation	Period
SB134	Bone	Hornfels	Inside	Buried	One year
SB135	Bone	Hornfels	Inside	Buried	Four weeks
SB136	Bone	Hornfels	Inside	Buried	Four weeks
SB137	Bone	Hornfels	Outside	Exposed	One year
SB138	Bone	Hornfels	Outside	Exposed	One year
SB139	Bone	Hornfels	Outside	Exposed	Four weeks
SB140	Bone	Hornfels	Outside	Exposed	Four weeks
SB141	Bone	Hornfels	Outside	Buried	One year
SB142	Bone	Hornfels	Outside	Buried	One year
SB143	Bone	Hornfels	Outside	Buried	Four weeks
SB144	Bone	Hornfels	Outside	Buried	Four weeks
SB145	Blank	Hornfels	Inside	Exposed	One year
SB146	Blank	Hornfels	Inside	Exposed	One year
SB147	Blank	Hornfels	Inside	Exposed	Four weeks
SB148	Blank	Hornfels	Inside	Exposed	Four weeks
SB149	Blank	Hornfels	Inside	Buried	One year
SB150	Blank	Hornfels	Inside	Buried	One year
SB151	Blank	Hornfels	Inside	Buried	Four weeks
SB152	Blank	Hornfels	Inside	Buried	Four weeks
SB153	Blank	Hornfels	Outside	Exposed	One year
SB154	Blank	Hornfels	Outside	Exposed	One year
SB155	Blank	Hornfels	Outside	Exposed	Four weeks
SB156	Blank	Hornfels	Outside	Exposed	Four weeks
SB157	Blank	Hornfels	Outside	Buried	One year
SB158	Blank	Hornfels	Outside	Buried	One year
SB159	Blank	Hornfels	Outside	Buried	Four weeks
SB160	Blank	Hornfels	Outside	Buried	Four weeks

A selection of 30 artefacts was collected from several Howiesons Poort layers (see Table 2.4). These included flakes and eight *pièces esquillées*. The tools are made on a variety ofrocks, ranging from hornfels to quartz. The term *pièces esquillées* is often used interchangeable with *outil ecaillés*, scaled pieces and core-reduced pieces. These artefacts are heavily flaked bifacial pieces with two opposing wide and straight crushed sides. It is unclear if they are by products, for example bipolar cores with two crushed striking platforms or tools with a specific function, such as a chisel (Thackeray 1981, Deacon 1984, Barham 1987). The subdivision of *outil ecaillés*, *pièces esquillées*, scaled pieces and core-reduced pieces is unclear and although some analysts make a technological distinction (Barham 1987), I use the terms *pièces esquillées* in this study.

This part of the study tests if there is a correlation in the preservation of ancient residues and the experimental residues. In addition, I test if residues on an assemblage of similar tool types have a comparable distribution pattern. I discuss the importance of distribution pattern below.

Williamson (2004, 2005) and Lombard (2004, 2005, 2006a, 2006b, 2008) conducted residue analysis on stone tools from Sibudu Cave. Both found plant, animal and inorganic (*e.g.* ochre) remains on artefacts from different stratigraphic layers. Lombard used residue analysis to demonstrate hafting of Howiesons Poort segments and post-Howiesons Poort points (Lombard 2006a, Lombard 2006b). The archaeological sample that I analysed has no overlap with previous studies.

The archaeological tools from Sibudu were specifically collected for this analysis. The tools were only handled by the excavator and immediately stored in plastic ziplock bags and brought to the University of the Witwatersrand for analysis. The tools were not washed and only handled by myself.

Table 2.4: Overview of the archaeological sample from Sibudu Cave.

Tool Number	Stone	Type	Level
SBL1	*Pièce esquillé*	Hornfels	Grey Rocky II
SBL2	Flake	Quartz	Grey Rocky II
SBL3	Flake	Quartz	Grey Rocky II
SBL4	Flake	Dolerite	Grey Rocky II
SBL5	Flake	Dolerite	Grey Rocky II
SBL6	Flake	Dolerite	Grey Rocky II
SBL7	Flake	Dolerite	Grey Rocky II
SBL8	Broken flake	Dolerite	Grey Rocky II
SBL9	*Pièce esquillé*	Quartz	Grey Rocky
SBL10	*Pièce esquillé*	Hornfels	Grey Rocky
SBL11	Flake	Quartz	Grey Sand
SBL12	*Pièce esquillé*	Quartz	Grey Sand
SBL13	Pointed flake	Dolerite	Grey Sand
SBL14	*Pièce esquillé*	Hornfels	Grey Sand
SBL15	Broken flake	Dolerite	Grey Sand
SBL16	Broken flake	Hornfels	Grey Sand
SBL17	Flake	Hornfels	Grey Sand
SBL18	Pointed flake	Dolerite	Grey Sand
SBL19	Flake	Dolerite	Grey Sand
SBL20	Flake	Dolerite	Grey Sand
SBL21	Flake	Quartz	Grey Sand
SBL22	Flake	Dolerite	Grey Sand
SBL23	*Pièce esquillé*	Dolerite	Grey Sand II
SBL24	Flake	Dolerite	Pinkish Grey Sand
SBL25	Flake	Hornfels	Pinkish Grey Sand
SBL26	*Pièce esquillé*	Dolerite	Pinkish Grey Sand
SBL27	*Pièce esquillé*	Quartz	Hearth I in Grey Rocky
SBL28	Flake	Dolerite	Hearth IV in GS
SBL29	Flake	Dolerite	Hearth IV in GS
SBL30	Flake	Hornfels	Hearth II in GS

2.1.3 Wilhelmina Polder and Zelhem sample

The Dutch experiments stand somewhat apart from the previously discussed samples. In this part of the project I want to investigate the influence of soil- and rainwater, soil types, stone grain sizes and microenvironments on residue preservation. These experiments are also not compared to an archaeological sample. Therefore, the design is different when compared to the South African experiments. The contrasting environments in the Netherland and South Africa in terms of precipitation may provide a useful and interesting comparison.

This sample consists of two locations in the Netherlands, Wilhelmina Polder and Zelhem (Figure 2.4). The two sites are used because the environments contrast greatly with the South African sites. Both Dutch locations are open-air sites with high annual precipitation. The sediments are more fertile compared to the Sibudu and Sterkfontein sediments. Wilhelmina Polder is located in the southwest of the Netherlands, in Zeeland province. The sediment at Wilhelmina Polder consists of marine clays. Zelhem is located in the east of the Netherlands, in Gelderland province. The sediment at Zelhem is sandy.

At both locations the following four identical experiments, unless stated otherwise, were set out. Thirteen flint tools were buried under approximately 15-20 centimetres of sediment and 13 tools were left at the surface. The exposed tools at Zelhem were deposited on the sand, but under the organic surface layer. Because there is no loose top layer at Wilhelmina Polder, the tools here were deposited on top of the soil and directly exposed to the elements. This part of the experiment tests whether there is a preservation difference between buried and exposed samples and if sand and clay soils show different preservation. It also tests whether there is a preservation bias between the residue types.

The experimental tools not were knapped specifically for this project, but selected from stones from knapping experiments at Leiden University. To avoid contamination, the tools were washed and dried prior to the experiments and handled with care. Flint was used for the tools because it is comparable in grain size to chert and hornfels. Before deposition, the flint tools were used to work extremely fatty bone and to cut potato, consequently creating fatty bone, bone, fat and starch residues on these tools. A blank sample was also deposited in the sediment and another on top. Again the blank sample is used as a reference and control sample for the residue

analysis on the used tools. Table 2.5 present the flint experiments at Wilhelmina Polder and Table 2.6 presents the flint experiments at Zelhem.

Table 2.5: Overview of the flint experiments at Wilhelmina Polder.

Tool number	Raw material	Worked material	Situation
NLC1	Flint	Starch	Exposed
NLC2	Flint	Starch	Exposed
NLC3	Flint	Starch	Exposed
NLC4	Flint	Starch	Exposed
NLC5	Flint	Starch	Exposed
NLC6	Flint	Starch	Buried
NLC7	Flint	Starch	Buried
NLC8	Flint	Starch	Buried
NLC9	Flint	Starch	Buried
NLC10	Flint	Starch	Buried
NLC11	Flint	Marrow/bone	Exposed
NLC12	Flint	Marrow/bone	Exposed
NLC13	Flint	Marrow/bone	Exposed
NLC14	Flint	Marrow/bone	Exposed
NLC15	Flint	Marrow/bone	Exposed
NLC16	Flint	Marrow/bone	Buried
NLC17	Flint	Marrow/bone	Buried
NLC18	Flint	Marrow/bone	Buried
NLC19	Flint	Marrow/bone	Buried
NLC20	Flint	Marrow/bone	Buried
NLC21	Flint	None	Exposed
NLC22	Flint	None	Exposed
NLC23	Flint	None	Buried
NLC24	Flint	None	Buried
NLC25	Flint	None	Buried

Table 2.6: Overview of the flint experiments at Zelhem.

Tool number	Raw material	Worked material	Situation
NLS1	Flint	Starch	Exposed
NLS2	Flint	Starch	Exposed
NLS3	Flint	Starch	Exposed
NLS4	Flint	Starch	Exposed
NLS5	Flint	Starch	Exposed
NLS6	Flint	Starch	Buried
NLS7	Flint	Starch	Buried
NLS8	Flint	Starch	Buried
NLS9	Flint	Starch	Buried
NLS10	Flint	Starch	Buried
NLS11	Flint	Marrow/bone	Exposed
NLS12	Flint	Marrow/bone	Exposed
NLS13	Flint	Marrow/bone	Exposed
NLS14	Flint	Marrow/bone	Exposed
NLS15	Flint	Marrow/bone	Exposed
NLS16	Flint	Marrow/bone	Buried
NLS17	Flint	Marrow/bone	Buried
NLS18	Flint	Marrow/bone	Buried
NLS19	Flint	Marrow/bone	Buried
NLS20	Flint	Marrow/bone	Buried
NLS21	Flint	None	Exposed
NLS22	Flint	None	Exposed
NLS23	Flint	None	Buried
NLS24	Flint	None	Buried
NLS25	Flint	None	Buried

The next set of experiments consists of glass microscope slides. On 22 slides small-grained sandpaper was glued. On 22 slides medium-grained sandpaper was glued. On 22 slides coarse-grained sandpaper was glued and 22 glass slides do not contain sandpaper. On the slides fat, blood and starch residues were deposited and 16 slides contained no residues. These blanks are used as a control sample. At the two sites 56 slides were buried, with the residues facing down and 24 slides were buried with the residues facing up. Tables 2.7 and 2.8 present the sandpaper experiments at Wilhelmina Polder and Zelhem respectively. This part of the experiments tests if there is differential residue preservation between raw materials with a different grain size. In addition, it tests if there is a preservation bias for tool sides facing up and down and if different residue types preserve similarly or not. If protective micro-environments influence residue preservation, the latter part could potentially demonstrate this.

Table 2.7: Overview of the sandpaper slide experiments at Wilhelmina Polder. Sandpaper S= small grained sandpaper; Sandpaper M= medium grained sandpaper; Sandpaper L= large grained sandpaper.

Tool number	Raw material	Worked material	Situation
NLCS1	Glass	Starch	Up
NLCS2	Glass	Starch	Up
NLCS3	Glass	Starch	Down
NLCS4	Glass	Starch	Down
NLCS5	Glass	Starch	Down
NLCS6	Glass	Starch	Down
NLCS7	Sandpaper S	Starch	Up
NLCS8	Sandpaper S	Starch	Up
NLCS9	Sandpaper S	Starch	Down
NLCS10	Sandpaper S	Starch	Down
NLCS11	Sandpaper S	Starch	Down
NLCS12	Sandpaper S	Starch	Down
NLCS13	Sandpaper M	Starch	Up
NLCS14	Sandpaper M	Starch	Up
NLCS15	Sandpaper M	Starch	Down
NLCS16	Sandpaper M	Starch	Down
NLCS17	Sandpaper M	Starch	Down
NLCS18	Sandpaper M	Starch	Down
NLCS19	Sandpaper L	Starch	Up
NLCS20	Sandpaper L	Starch	Up
NLCS21	Sandpaper L	Starch	Down
NLCS22	Sandpaper L	Starch	Down
NLCS23	Sandpaper L	Starch	Down
NLCS24	Sandpaper L	Starch	Down
NLCBL1	Glass	Blood	Up
NLCBL2	Glass	Blood	Up
NLCBL3	Glass	Blood	Down
NLCBL4	Glass	Blood	Down
NLCBL5	Glass	Blood	Down
NLCBL6	Glass	Blood	Down
NLCBL7	Sandpaper S	Blood	Up
NLCBL8	Sandpaper S	Blood	Up
NLCBL9	Sandpaper S	Blood	Down
NLCBL10	Sandpaper S	Blood	Down
NLCBL11	Sandpaper S	Blood	Down
NLCBL12	Sandpaper S	Blood	Down
NLCBL13	Sandpaper M	Blood	Up
NLCBL14	Sandpaper M	Blood	Up
NLCBL15	Sandpaper M	Blood	Down
NLCBL16	Sandpaper M	Blood	Down
NLCBL17	Sandpaper M	Blood	Down
NLCBL18	Sandpaper M	Blood	Down
NLCBL19	Sandpaper L	Blood	Up
NLCBL20	Sandpaper L	Blood	Up
NLCBL21	Sandpaper L	Blood	Down
NLCBL22	Sandpaper L	Blood	Down
NLCBL23	Sandpaper L	Blood	Down
NLCBL24	Sandpaper L	Blood	Down
NLCF1	Glass	Fat	Up

Continuation Table 2.7: Overview of the sandpaper slide experiments at Wilhelmina Polder.

NLCF2	Glass	Fat	Up
NLCF3	Glass	Fat	Down
NLCF4	Glass	Fat	Down
NLCF5	Glass	Fat	Down
NLCF6	Glass	Fat	Down
NLCF7	Sandpaper S	Fat	Up
NLCF8	Sandpaper S	Fat	Up
NLCF9	Sandpaper S	Fat	Down
NLCF10	Sandpaper S	Fat	Down
NLCF11	Sandpaper S	Fat	Down
NLCF12	Sandpaper S	Fat	Down
NLCF13	Sandpaper M	Fat	Up
NLCF14	Sandpaper M	Fat	Up
NLCF15	Sandpaper M	Fat	Down
NLCF16	Sandpaper M	Fat	Down
NLCF17	Sandpaper M	Fat	Down
NLCF18	Sandpaper M	Fat	Down
NLCF19	Sandpaper L	Fat	Up
NLCF20	Sandpaper L	Fat	Up
NLCF21	Sandpaper L	Fat	Down
NLCF22	Sandpaper L	Fat	Down
NLCF23	Sandpaper L	Fat	Down
NLCF24	Sandpaper L	Fat	Down
NLCB1	Glass	None	Up
NLCB2	Glass	None	Up
NLCB3	Glass	None	Down
NLCB4	Glass	None	Down
NLCB5	Sandpaper S	None	Up
NLCB6	Sandpaper S	None	Up
NLCB7	Sandpaper S	None	Down
NLCB8	Sandpaper S	None	Down
NLCB9	Sandpaper M	None	Up
NLCB10	Sandpaper M	None	Up
NLCB11	Sandpaper M	None	Down
NLCB12	Sandpaper M	None	Down
NLCB13	Sandpaper L	None	Up
NLCB14	Sandpaper L	None	Up
NLCB15	Sandpaper L	None	Down
NLCB16	Sandpaper L	None	Down

Table 2.8: Overview of the sandpaper experiments at Zelhem. Sandpaper S= small grained sandpaper; Sandpaper M= medium grained sandpaper; Sandpaper L= large grained sandpaper.

Tool number	Raw material	Worked material	Situation
NLSS1	Glass	Starch	Up
NLSS2	Glass	Starch	Up
NLSS3	Glass	Starch	Down
NLSS4	Glass	Starch	Down
NLSS5	Glass	Starch	Down
NLSS6	Glass	Starch	Down
NLSS7	Sandpaper S	Starch	Up
NLSS8	Sandpaper S	Starch	Up
NLSS9	Sandpaper S	Starch	Down
NLSS10	Sandpaper S	Starch	Down
NLSS11	Sandpaper S	Starch	Down
NLSS12	Sandpaper S	Starch	Down
NLSS13	Sandpaper M	Starch	Up
NLSS14	Sandpaper M	Starch	Up
NLSS15	Sandpaper M	Starch	Down
NLSS16	Sandpaper M	Starch	Down
NLSS17	Sandpaper M	Starch	Down
NLSS18	Sandpaper M	Starch	Down
NLSS19	Sandpaper L	Starch	Up
NLSS20	Sandpaper L	Starch	Up
NLSS21	Sandpaper L	Starch	Down
NLSS22	Sandpaper L	Starch	Down
NLSS23	Sandpaper L	Starch	Down
NLSS24	Sandpaper L	Starch	Down
NLSBL1	Glass	Blood	Up
NLSBL2	Glass	Blood	Up
NLSBL3	Glass	Blood	Down
NLSBL4	Glass	Blood	Down
NLSBL5	Glass	Blood	Down
NLSBL6	Glass	Blood	Down
NLSBL7	Sandpaper S	Blood	Up
NLSBL8	Sandpaper S	Blood	Up
NLSBL9	Sandpaper S	Blood	Down
NLSBL10	Sandpaper S	Blood	Down
NLSBL11	Sandpaper S	Blood	Down
NLSBL12	Sandpaper S	Blood	Down
NLSBL13	Sandpaper M	Blood	Up
NLSBL14	Sandpaper M	Blood	Up
NLSBL15	Sandpaper M	Blood	Down
NLSBL16	Sandpaper M	Blood	Down
NLSBL17	Sandpaper M	Blood	Down
NLSBL18	Sandpaper M	Blood	Down
NLSBL19	Sandpaper L	Blood	Up
NLSBL20	Sandpaper L	Blood	Up
NLSBL21	Sandpaper L	Blood	Down
NLSBL22	Sandpaper L	Blood	Down
NLSBL23	Sandpaper L	Blood	Down
NLSBL24	Sandpaper L	Blood	Down
NLSF1	Glass	Fat	Up
NLSF2	Glass	Fat	Up
NLSF3	Glass	Fat	Down
NLSF4	Glass	Fat	Down

Continuation Table 2.8: Overview of the sandpaper experiments at Zelhem.

Tool number	Raw material	Worked material	Situation
NLSF5	Glass	Fat	Down
NLSF6	Glass	Fat	Down
NLSF7	Sandpaper S	Fat	Up
NLSF8	Sandpaper S	Fat	Up
NLSF9	Sandpaper S	Fat	Down
NLSF10	Sandpaper S	Fat	Down
NLSF11	Sandpaper S	Fat	Down
NLSF12	Sandpaper S	Fat	Down
NLSF13	Sandpaper M	Fat	Up
NLSF14	Sandpaper M	Fat	Up
NLSF15	Sandpaper M	Fat	Down
NLSF16	Sandpaper M	Fat	Down
NLSF17	Sandpaper M	Fat	Down
NLSF18	Sandpaper M	Fat	Down
NLSF19	Sandpaper L	Fat	Up
NLSF20	Sandpaper L	Fat	Up
NLSF21	Sandpaper L	Fat	Down
NLSF22	Sandpaper L	Fat	Down
NLSF23	Sandpaper L	Fat	Down
NLSF24	Sandpaper L	Fat	Down
NLSB1	Glass	None	Up
NLSB2	Glass	None	Up
NLSB3	Glass	None	Down
NLSB4	Glass	None	Down
NLSB5	Sandpaper S	None	Up
NLSB6	Sandpaper S	None	Up
NLSB7	Sandpaper S	None	Down
NLSB8	Sandpaper S	None	Down
NLSB9	Sandpaper M	None	Up
NLSB10	Sandpaper M	None	Up
NLSB11	Sandpaper M	None	Down
NLSB12	Sandpaper M	None	Down
NLSB13	Sandpaper L	None	Up
NLSB14	Sandpaper L	None	Up
NLSB15	Sandpaper L	None	Down
NLSB16	Sandpaper L	None	Down

The experimental flint tools and slides from Zelhem and Wilhelmina Polder were carefully collected after six months. The samples were stored in plastic ziplock bags. They were not washed prior to analysis and only handled by myself, avoiding post-excavational contamination.

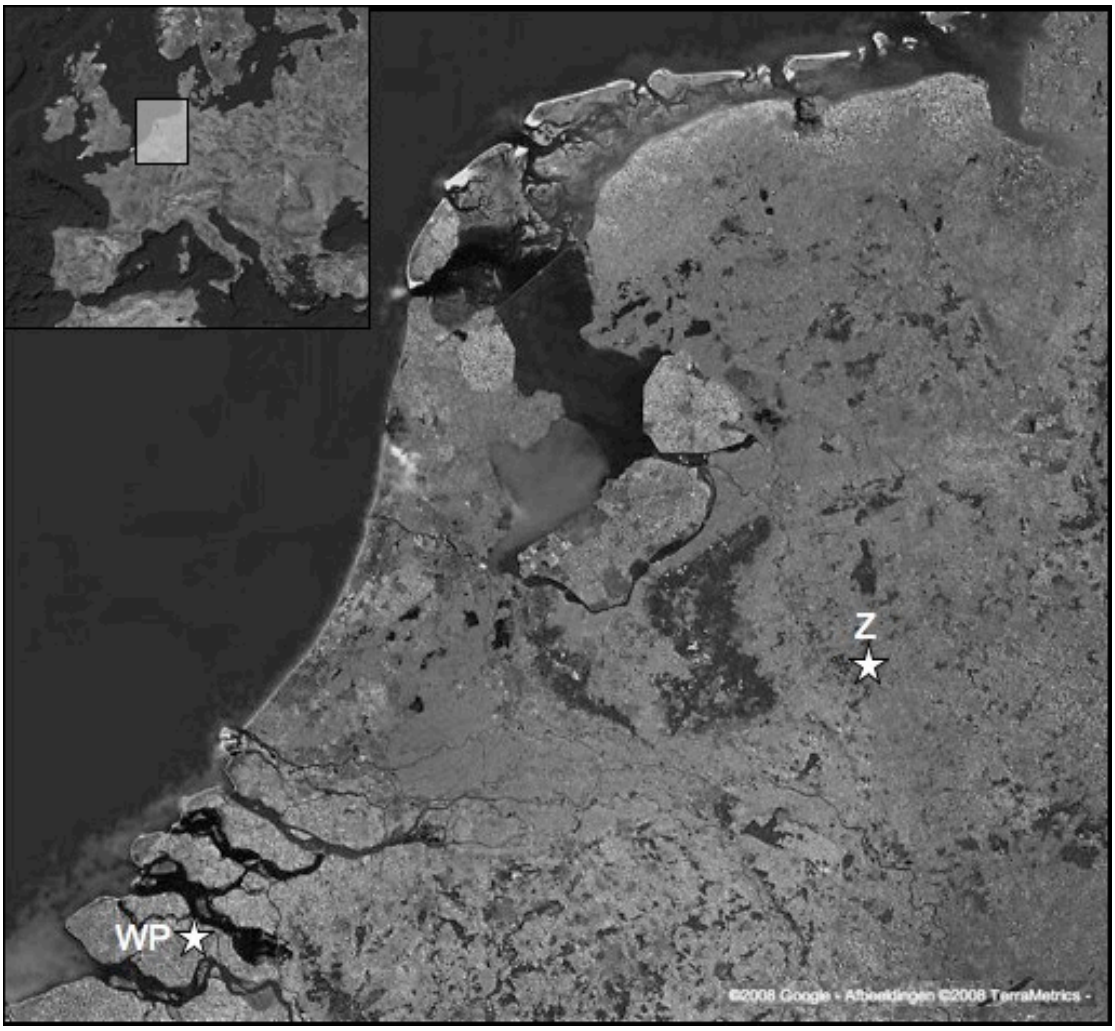


Figure 2.4: Map with the location of the two Dutch sites. WP= Wilhelmina Polder; Z= Zelhem (www.googlemaps.com 2008).

2.2 ANALYSIS

In the following paragraphs and chapters I use the term use-related residue. These are residues that are ancient and intentional and only these residues are useful in residue analysis. Old post-depositional and modern post-excavational contaminants also occur on the experimental and archaeological tools. In the following chapters I use the term contaminant for sediment organic remains and sometimes also for ancient unintentional and not use-related residues.

The term "invasive" in Chapters Four to Six is used to indicate the location of residues on the tool's surface. Invasive residues are present in the middle or towards

the interior of a tool. Contrasting invasive residues are residues that are confined to the working edge. The two conditions are used to describe distribution patterns of residue types, such as bone or muscle tissue, and contaminants.

Many worked experimental tools were covered in (compact) sediment. The damp residues served as an adhesive for the sediment. When archaeological tools also show signs of sediment adhering to them I use this as an indication of possible processing with the tools in the past.

2.2.1 Microscopy

The tools were analysed with an indirect or reflected light microscope (Olympus BX30) at magnifications ranging from 50 to 800. The individual residues were photographed with an Olympus digital camera. See the figures in Appendix B for examples of micrographs of residues. Part of the sediment samples and the entire glass sample in the sandpaper experiments were analysed under a direct or transmitted light microscope (Olympus BX40). However, pictures of these samples were taken under the indirect light microscope. The micrographs aid the identification of residues and are used to illustrate residues. Detailed maps, recording the distributions of residues, were compiled per ventral and dorsal side of a tool. See the figures in Appendix B for example of distribution maps. The distribution maps help to distinguish meaningful patterns, which might appear on used tools and the maps help to reveal if this is different from the pattern of organic sediment remains on the blank flakes. The distribution maps are the means through which I apply a contextual approach to analyse residues.

A reflected light microscope was selected for my study. Some residue researchers, starch analysts in particular, prefer the use of transmitted light microscopes (*e.g.* (Barton, Torrence and Fullagar 1998, Giovannetti *et al.* 2008, Horrocks *et al.* 2008, Lentfer, Therin and Torrence 2002, Piperno *et al.* 2004). Transmitted light microscopy is used on transparent samples as the light is directed from below through the sample. Reflected light microscopy is for dense samples and the light is directed from above onto the subject. Because stone tools are solid they can only be analysed with a reflected light microscope, unless the residues are physically removed from the

tools. For starch analysis, but potentially for all residue types, the residues can be extracted from the tools using deionized water and a pipette. The residues are then placed on microscope slides and are studied with a transmitted light microscope. The advantage of a transmitted light microscope is that the images are much clearer than from a reflected light microscope. The structure of the residues, such as cells and cell walls, are better visible and light properties, such as birefringency, are clearer. Additionally, the extracted samples can be stained and worked with chemicals to achieve even better visibility. Differential Interference Contrast microscopy (DIC) samples allow for accurate analysis, which is useful when visual characteristics are used for taxonomic identification of starch grains (*e.g.*Fullagar *et al.* 2006). There are occasions when the use of transmitted light microscopy is the only option, for example, in the study of rock art panels, where small paint samples have to be extracted (*e.g.* Williamson 2000a).

Although transmitted light microscopy appears superior to reflected light microscopy, there are some disadvantages. The residues are extracted from the tools, so the method is destructive and future analysts cannot view the residue in its original context. Individual residues are dissolved in water and pipetted from the surface to a microscope slide. In many studies the whole tool is eventually or initially placed in a (sonic) bath to extract all residues (*e.g.* Fullagar *et al.* 2006, Perry 2004, Piperno *et al.* 2004). The first technique is time consuming and when analysing large samples it is impossible to extract even half of the encountered residues. With the second method, the context of the residues on the tool' surface is lost and, as is explained below, it is impossible to distinguish contaminants from use-related residues. Because I analysed large samples for this study and because the context of the residues is vital, I preferred using a reflected light microscope.

2.2.2 The contextual approach and identification of contaminants

One major concern regarding residue analysis is the identification of post-depositional and post-excavational contaminants. Williamson (Rots and Williamson 2004, Williamson 2006) found that starch grains from the sediment and nearby tubers can adhere to stone tools. Airborne starch grains can also contaminate tools (Hinojosa 2000, Barton and Matthews 2006, Loy and Barton 2006). Barton *et al.* (Barton, Torrence and Fullagar 1998) found that although starch from sediments adheres to

tools, use-related starch remains are more abundant. Rots and Williamson (2004) also note that contaminants are more isolated and that use-related residues form a matrix. Williamson (2006) demonstrates that tubers in the sediment can contaminate a tool with starch in large quantities, similar to use-related quantities. Another contaminant group is the unintentional, but ancient micro-remains. These remains are from handling by ancient toolmakers. Post-excavational handling can contaminate tools with modern residues, for example starch grains in surgical gloves, plant fibres from paper storage bags, or ink from dirty hands (Wadley and Lombard 2007, Wadley pers. comm. 2007, Lombard pers. com. 2007, 2008). Some contaminants are easily distinguished. Bright blue cotton fibres and pink ink, for example, are obvious modern residues. Other remains appear similar to use-related residues. Ancient contaminants are sometimes difficult to discern from use-related residues. Because they also have been buried for a long period, they appear similar to use-related residues. I found many fatty residues and some resin, which result from unintentional handling by prehistoric people. Because these are the same types as use-related residues, differentiation is complex.

In this study I apply the contextual approach, following Lombard and Wadley (Lombard and Wadley 2007, Wadley and Lombard 2007), to residue analysis to discriminate use-related residues from contaminants (Figure 4.10). Association and distribution are the parameters for this approach: association between residues (number 2 in Figure 4.10) and association between residues and their positioning on a tool (number 3 in Figure 4.10). These two parameters can be used to demonstrate the authenticity of residues. The third parameter, the distribution of all residues on a tool (number 4 in Figure 4.10), is used to reconstruct the processed materials. Consequently, single residues with an illogical distribution are ambiguous and the study of residue suites is ideal for processed material identification.

The total context of all remains, the possible use-related residues and contaminants, on a tool is important. For example, graphite, nylon fibres and nail varnish all indicate contamination of a tool. If starch grains are also present on the same tool, and even if they occur on a thin and possible working edge, they are ambiguous remains. By analysing suites of contaminants it is possible to eliminate them successfully from an analysis. The same comment is applicable for use-related residues, which are most convincingly identified as suites of related residues.

On a sample level, meaningful patterns must be repeated to indicate tool use for a tool type. Not all tools in a selected sample always show a distinct pattern, but a majority of the sample must exhibit repeated patterns to permit generalisations. Consequently, the analysis of a random sample of artefact types is ineffectual. No meaningful patterns appear at such a sample level and it is difficult to distinguish reoccurring contaminants from use-related patterns. The analysis of single artefacts or small samples is equally ineffectual, unless agency is applied (see Chapter One).

The identification of contaminants is based on the contextual approach, explained above. Chapter Four demonstrates that sediment organics have a random spread over the tool surface and that use-related residues show meaningful distribution patterns. All samples in this analysis contained contaminants. Contaminants can come from handling or storage and from the sediments in which artefacts were buried. It is important to know what the possible contaminants are in order to exclude them from the analysis. Therefore I examined unused blank flakes, which were deposited at the sites and sediment samples. Identification criteria for contaminants such as hyphae and pollen are discussed below. The sediment samples are discussed per site in Chapter Three.

Sediment samples must be analysed to understand what is available in the sediment matrix. If sediment contains many plant fibres and plant tissue one needs to be extra careful with the identification of ancient plant remains on tools. Sediment contaminants can look quite different from faded archaeological use-related residues, as the latter have probably been exposed longer.

Sediment patches and layers on the artefact are useful. They can serve as a "stratigraphic base layer", meaning that organic remains that lay on top of these patches were deposited after the sediment layer and after the artefact was deposited in the sediment and hence they are contaminants.

Contaminants are similar on all artefacts from the same assemblage. Consequently, a selection of random artefacts, belonging to different types, gives a good overview of contaminants in a particular occupation of the same site. If a particular residue is present on a large percentage of a random sample, the residue is most likely a contaminant. On the other hand, a reoccurring residue on a sample of

similar tool types is part of a meaningful pattern and indicates that the residue is probably use-related.

Organic micro-remains often look different on stone than on a sediment sample slide, which is generally used for analysis. On stone, light refracts differently and the colour of the stone also influences the way contaminants and residues look. It is useful to set out blanks around the excavation a few months before the start of the residue study. These blanks form an important part of a reference collection as they help with the identification of contaminants and with the raw material investigation. In addition, it is helpful to look at sediment patches on the archaeological sample. Archaeological tools can contain different contaminants from those on the modern blanks and these can be discriminated and viewed on sediment patches.

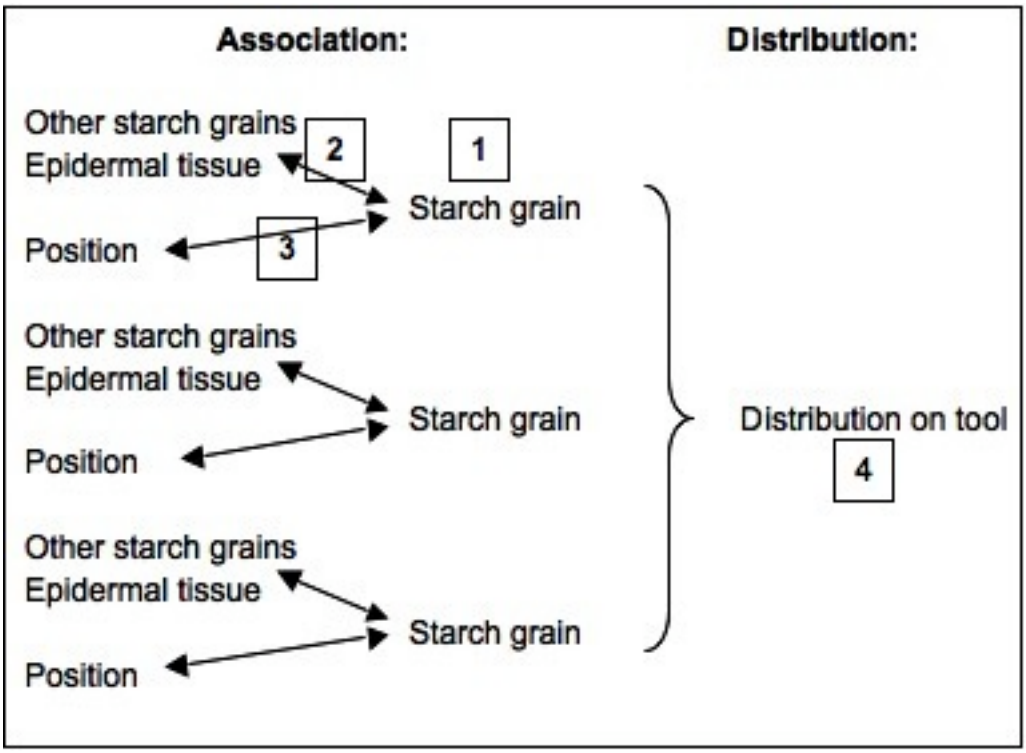


Figure 2.5: Example of the contextual approach. 1: identification of a residue. 2: identification of associated residues. 3: study into the positioning of the single and related residues. 4: study into the distribution of all the remains on a tool.

2.2.3 Stone surface

The stone surface of a tool greatly influences the identification of residues. Reflectiveness, colour, mineral inclusion and homogeneity are properties that

determine if a rock type allows for straightforward analysis or not. The more reflective the stone, the more difficult it is to see the residue and determine its characteristics. Dark stone is best for analysis. On the one hand, dark colours contrast with the residues and are easier on one's eyes. Quartz, on the other hand, is so reflective and bright white that residues are difficult to spot (Figure A.1). Mineral inclusions can mimic residues. Mineral inclusions also break the light differently, obscuring contaminants. An even, homogeneous background makes analysis much easier because there are fewer reoccurring mineral inclusions in such rocks. For example, a cortical surface is difficult to analyse (Figure A.2). Before commencing residue analysis I suggest one investigates the rock types. Understanding the stone and its properties will save time once the analysis of the samples starts. Fracturing can mimic residues and in paragraph 2.3.4 I discuss breaking patterns and micro-wear in more detail.

In these experiments I used hornfels for the Sibudu experiments, chert for the Sterkfontein experiments and flint and also red sandpaper for the clay and sand experiments. The grain sizes from the first three are comparible. The light grey Meuse flint from the south of the Netherlands, used here, has very fatty and sometimes reflective properties, obscuring and mimicking residues (Figure A.3). Damaged flint appears like bone (Figure A.90). However, Meuse flint is homogeneous and it does not contain many inclusions.

Chert is more reflective, but only has small uniform inclusions (Figure A.4). The chert used in these experiments varies between quite dark grey, which works well under the microscope, to light grey, which is to harder to work with. Chert is similar to flint and breaks also mimic bone residues (Figure A.91), but the material used here does not appear as fatty.

Black hornfels allows for easy viewing of the residues because these contrast well against the dark background (Figure A.5). Hornfels is not very reflective and it is homogeneous and does not have large inclusions.

The red sandpaper is obviously homogeneous; it is not too reflective and does not have any inclusions (Figures B.133-B.135). Its bright and dark red colour makes an easy contrast to the contaminants and residues.

In general, dark homogeneous stone is easier for analysis. However, stone materials can create a bias in the analysis. For example, on black hornfels, light starch

deposits are easily identified, whereas they are difficult to see on quartz. Stains and muscle tissue, on the other hand, are very clear on quartz, but almost impossible to see on the dark hornfels. Consequently, muscle tissue and stains are probably underrepresented on dark stone. In Chapter 6, I discuss the results from the Sibudu archaeological sample and I address this issue in more detail.

2.2.4 Preservation index

In order to cross-examine preservation between replicated tools and sites, the experimental residues required some form of quantification. Every experimental tool received a number indicating how much of the original residue was left. I prepared drawings of every tool on graph paper, allowing me to count the residues in a one by one millimetre grid. However, it proved impossible to count all residues and to design a workable index. Instead I used the detailed distribution maps to create a more descriptive index as presented in Table 2.9.

Working on a starchy material leaves many residues on a tool, but bone and wood leave only few residues. The index takes into account that the initial stages of the production of some residues are abundant whereas this is not the case for production of other residues. Hardy *et al.* (2001) also note that hard processed materials leave fewer residues on the working edge of the tool than softer materials. In the design of the index, a residue type was only compared to itself and its initial stage of production. This means that the quantity of residues present at an index of 3 for starch and bone tools can be different, but also that these residues are at the same stage of deterioration. Consequently, the preservation index allows cross-comparison of residue types.

Although it is theoretically impossible to score a 5, which represents the situation directly after use, I used this score to indicate exceptional preservation. In the scoring of starch samples I only analysed the preservation of the starch grains. Although in many cases epidermal tissue is preserved, these experiments were set up to investigate the preservation of particular materials, not to reconstruct tool use. In the index, however, I do mention epidermal tissue to show how, as one residue declines, another becomes more dominant. Fat is impossible to separate from bone residues and therefore the two are linked in the index. Most residues form recognisable combinations, for example, muscle tissue, fat and bone often come

together, but I tried to apply the index only on residues that I was investigating. This means that many tools with starch residues also contain epidermal tissue, but the epidermal tissue is not used in the index.

I did not apply the same preservation index to the sandpaper experiments described in paragraph 2.1.3. The samples were too small to be compared to the flakes used in the other experiments. The sandpaper pieces were not used as tools; instead the residues were applied on them. The sandpaper index is as follows: (2) many residues are present (1) few residues are present (0) no residues are present.

Table 2.9: Preservation index

	Preservation index					
Residue type	5	4	3	2	1	0
Starch	Situation just after use. Thick deposits on the working edges, but also clusters of starch grains all over the surface. Epidermal tissue present along the edge and in interior, but not important for identification	Abundant presence of starch in thick deposits along the working edge, but also in interior of tool. Epidermal tissue present along the edge and in interior, but not important for identification.	Along the edge deposits present, but not abundant. In interior mainly single grains and small clusters.	Few deposits left. Mainly single grains or small clusters in combination with epidermal tissue. Largest concentrations along working edge	The occasional residue left. Often only epidermal tissue and few single grains	No observed residue left
Plant tissue	Situation just after use. Thick deposits of wood fibres and plant tissue concentrated along the working edges, but also invasive.	Abundant presence of wood tissue and fibres mainly along edge.	Along the edge deposits present, but not abundant.	Few deposits left. Mainly small macerated deposits and single fibres. Largest concentrations along working edge	The occasional residue left. Small deposits of macerated tissue or small fibres. Often only distinguishable due to location on working edge.	No observed residue left
Bone	Situation just after use. Thick bone deposits, which are only concentrated along the working edge. Fat can be more invasive, but rarely.	Relative abundance of bone flakes and deposit along the edge. Fat always present in combination with bone and is more widespread.	Deposits present along the edge, but not abundant. Fat is relatively more dominant.	Very few deposits left. Concentrations along the edge and dominated by fat and fatty bone.	The occasional residue left. Fat seems more persistent. Often only distinguishable due to location on working edge	No observed residue left
Muscle tissue	Situation just after use. Thick muscle tissue and blood stains are located along the working edge and residues can be quite invasive.	Abundant deposits of macerated tissue and striated muscle tissue along the working edge and towards interior. Some connective fibres.	Along the edge larger concentrations of tissue present, but not abundant. Some connective fibres.	Few deposits left concentration along edge, but also in interior. Connective fibres/bony tissue relatively more abundant.	The occasional residue left. Small deposits of striated muscle tissue in combination with fat are identifiable. Muscle tissue alone often resembles plant by this stage too much. Often only distinguishable due to location on working edge	No observed residue left
Fat	Situation just after use. Thick fat deposits present along working edge and more in interior.	Abundant deposits of fat cells and deposits along edge and towards interior.	Along the edge larger deposits present. By this stage often no cells visible anymore.	Few deposits left mainly along the working edge.	The occasional residue left. Sometimes only a dark shadow left on the surface. Often only distinguishable due to location on working edge	No observed residue left

2.3 IDENTIFICATION CRITERIA

Residue types are identified by characteristic visual cues. In the following paragraphs I discuss residue types and their identification criteria. Undoubtedly, more residues types than the ones discussed exist. However, a specialist draws only from his/her experience and so I can only discuss types, which I encountered myself. The residue types introduced below are those needed to understand this study (for further residue descriptions see Lombard 2004, Wadley, Lombard and Williamson 2004, Lombard 2005, Fullagar 2006).

2.3.1 Plant residues

Plant tissue and fibres

Plant cells have cell walls, which animal cells do not have. Cell walls are composed of cellulose fibres, which are embedded in hemicellulose and lignin and other polysaccharides and proteins (Campbell 1990). There are five main types of plant cells (Devlin and Witham 1983, Mauseth 1988, Campbell 1990).

1. Parenchyma cell (Figure A.6A): is a typical plant cell and it is the most unspecialised plant cell. These cells lack secondary walls and their primary wall remains thin and flexible. They usually have a large central vacuole, a storage membrane compartment, within the cell. Parenchyma is present in a wide variety of tissues.
2. Collenchyma cell Figure A.6B): usually lacks secondary walls, but their primary walls are thicker than parenchyma cells and also unevenly thickened. They are usually grouped in strands or cylinders in young, growing plant parts.
3. Sclerenchyma cell (Figure A.6C): has a thick secondary wall, strengthened by lignin. The cells are specialised for support and are much more rigid than collenchyma. Sclerenchyma occurs in plant parts that no longer grow. The cells are subdivided into fibres and sclereids. Fibres occur in bundles and are long, slender and tapered. Sclereids are shorter than fibres and irregular in form (Figure A.6D).
4. Water-conducting cell (Figure A.6E): is an elongated cell that facilitates water transport in plants and this type can be subdivided into tracheids and vessel

elements. Both elements have secondary walls, which in growing tissue are unevenly deposited in a ring or spiral pattern. When these cells complete their differentiation they die and leave behind an element through which water can flow. Tracheids are long and thin with tapered ends and vessels are wider and shorter with thinner cell walls. Water-conducting cells are part of xylem tissue systems.

5. Sieve-tube cell (Figure A.6F): transports nutrients. A chain of these cells form a tube through which sugars and ions are transported. They are very simple cells, which lack a nucleus, have few vacuoles and lack a secondary cell wall. The walls have pores to allow fluids to flow from cell to cell. Unlike water-conducting cells, sieve-tube cells are alive. Sieve-tube cells are part of phloem tissue systems.

Plant tissue consists of groups of cells, which are differentiated in simple and complex tissues. Simple tissue contains cells of one type, for example parenchyma, and is named after that cell type, for example parenchyma tissue. Complex tissues contain more than one type of cell. There are three complex tissue systems, namely: the dermal or epidermis, vascular and ground tissue systems. The epidermis consists of tightly packed cells, which form the outer surface and protection of leaves and young plant parts. Epidermal tissue consists of specialised parenchyma cells, which for example have hairs or secret waxy cuticle (Devlin and Witham 1983, Mauseth 1988, Campbell 1990).

The vascular system consists of xylem and phloem tissues, both are specialised in transport of water and nutrients respectively. Xylem contains water-conducting cells and generally also has sclerenchyma cells (fibres) for support and parenchyma cells for storage. Phloem contains sieve-tube cells and sclerenchyma cells (sclereids) and parenchyma cells (Devlin and Witham 1983, Mauseth 1988, Campbell 1990).

Ground tissue makes up the bulk of young plant and it is less differentiated than the other tissues. It fills the space between the vascular and dermal tissue systems and it predominantly contains parenchyma, but also some collenchyma and sclerenchyma cells. Ground tissue manufactures nutrients by photosynthesis and it stores (Devlin and Witham 1983, Mauseth 1988, Campbell 1990).

In general, plant tissue is recognised under the microscope by the presence of cell walls, large vacuoles and green chlorophyll cells. The rigid organisation of plant tissue gives plant matter a characteristic stiff and angular feel. Woody tissue contains characteristic elongated xylem cells (Figure A.7). Epidermal tissue has a rectangular brick like structure and the individual cells are easily distinguished (Figure A.8). Dermis cells are sometimes covered with cutin or a wax layer. Plant fibres are bundles of sclerenchyma fibre cells. The cells in plant fibres are long, slender and ribbon-like and, when torn, the ends of single strands in the bundle are twisted and appear shattered (Figures A.9 and A.10). The breaks in fibres are usually straight and abrupt. Plant tissue is birefringent (Figures A.8 and A.10), meaning that it causes light to refract and in doing so the surface of the plant residue becomes bright and reflective and shows rainbow colours. Plant tissue can also be translucent under high magnifications. Other plant residues such as starch grains, resin and exudate often accompany plant tissues and fibres.

Modern fibres are common contaminants on artefacts (Figure A.11). They have bright colours ranging from reflective white to blue. Post-depositional and post-excavational plant residues are usually extremely birefringent and they usually also have bright or fresh colours. Ancient residues tend to take on colours from the sediment and generally appear more faded than fresh resdiues.

Starch grains

Starch grains are specialised parenchyma cells in which plants store energy. They are most abundant in storage organs, such as roots, tubers and seeds, but can also be found in smaller quantities in leaves and other plant parts (Haslam 2004). Starch grains predominantly consist of two polysaccharides: (1), amylose and (2), amylopectin (Lillford and Morrison 1997). Inside the grains, glucose forms double and single helixes that create a crystalline structure (Gidley 2001). Under polarized light this crystalline structure produces the distinctive Maltese, or extinction cross (Gidley 2001). Concentric rings of amylose and amylopectin are formed as the granule develops. These growth rings are more pronounced in large grains, such as potato, and under hydrated conditions (Donald, Perry and Waigh 2001). They vary in size from 1 micrometre to 175 micrometre. The large grains are mainly present in storage organs and the smaller ones throughout the plant. Starch grains can be circular to ellipse and triangular shaped. The small, ever-present starch grains are generally

circular. Particular shapes of starch grains and extinction crosses are attributed to specific plant species and starch is used to, for example, reconstruct palaeodiets and subsistence (Reichert 1913, Czaja 1978, Loy 1994, Piperno and Holst 1998, Gott *et al.* 2006). Starch grains are birefringent and translucent, but damaged starch grains become opaque (Barton and Matthews 2006) personal observation, see Chapter Four).

Starch grains are also present in sediments. Usually they are quite small, between 2 and 5 micrometre. However, the sediments at Wilhelmina Polder and Zelhem in this study also contained larger examples, comparable to the potato starch (up to 50 micrometre). The majority of starch grains are only visible under a direct light microscope. It is therefore advisable to use a direct light microscope for all sediment samples. One needs to keep in mind that starch from sediments can easily be mistaken for use-related residues.

Degraded plant tissue and charcoal

Depending on degeneration of the plant tissue, a skeleton of cell walls and cell structures can become clearer. When microbes attack a plant cell they focus on the nutrients inside the cell, leaving the cellulose in the cell walls for specialised bacteria. Degraded plant tissue can be white or light yellow (Figures A.15-A.17), but usually it is dark brown in colour. Degraded wood is often fragmented.

Charcoal, burnt plant tissue, varies in colour between brown and black, depending on the temperature and burning process. The plant cells carbonise during burning and this leaves a carbon skeleton behind (Figure A.18). Degraded plant tissue and charcoal appear similar through the microscope. Dermal tissue, for example degraded leaf matter from the sediment, resembles a carbon skeleton, but the cells in epidermis are tightly packed and of similar size (A.19). The carbonised cells in charcoal usually originate from xylem or phloem tissue and the cells are larger and vary in size and less tightly packed. Burnt plant tissue can have a fatty gloss. Both degraded and burnt tissue can be birefringent (Figure A.19). However, in the Sibudu sediment samples, degraded wood also appeared white and in some cases the tissue is so deteriorated that it resembles (fatty) bone (Figures A.15 and A.17). In several cases the tissue was not birefringent.

Resin

Resin is a general term for plant secretions and is sometimes also confused with petroleum-based materials, such as bitumen. The term resin often includes pitch, (waxy) oils, gums and sometimes even latex. However, these plant secretions differ in composition and origin from each other (Mauseth 1988). Plant oils vary from volatile to waxy and they are deposited in cavities inside the plant. Oils can move outside the plant where they are aromatic (Mauseth 1988). Gum is found inside the wood of certain tree species (Mauseth 1988). Latex is a general term for creamy and thick secretions, which are often described as white and milky in appearance. However, latex can also be clear, yellow or orange.

In the literature it is often unclear what plant secretion a particular resin is. Connan and Nissenbaum (Connan and Nissenbaum 2003) recommend that pristine resin should be differentiated from tar and pitch. Pristine resins are not processed, whereas tar and pitch are the by-products of distillation of resinous wood (Connan and Nissenbaum 2003, Font *et al.* 2007). Pitch, tar and pristine resins are complex mixtures, but mainly contain (volatile) aromatic hydrocarbons or terpenes and resin acids, which are the main constituent of essential oils (Mauseth 1988). In addition, resin can contain salts, alkaloids, sterols, lipids, tannins, mucilages, camphors, rubber, proteins, vitamins, crystals, starch grains and even living protozoans (Mauseth 1988).

Tar is viscous black liquid derived from destructive distillation of organic matter, such coal or wood. The first produces coal tar and is similar to materials such as bitumen and asphalt. Wood tar is extracted by heating resinous wood like pinewood or birch bark. Tar bleeds from the wood, leaving a charcoal mass behind. Distilling liquid terpene plant secretions produces turpentine and pitch. Pitch, also known as rosin, is similar to tar, but more solid. It can shatter on impact, but it is actually an extremely slow flowing fluid (Mauseth 1988, Van Bergen *et al.* 1997, Connan and Nissenbaum 2003).

Unlike pitch and tar, some resins are simply collected and not processed. Frankincense, for example, is tapped from *Boswellia* sp. trees by slashing the bark and allowing the exudated resins to bleed out and harden (Van Bergen *et al.* 1997). These secretions are well known from conifer trees, but many other trees including *Acacia* sp. also produce pristine resin (Goffer 2007). Although *Acacia* sp. more precisely secretes a gum, (Figure A.20B) I make no further distinction in this thesis between

pristine resins. With the microscope it is not possible to distinguish different types of secretion and in addition, it confuses the terminology.

Resins are sometimes mixed with other resins, petroleum-based products, animal fat, beeswax or honey to create specific properties (Forrest 1982, Dudd and Evershed 1999, Evershed *et al.*2001, Regert 2004, Van Gijn and Boon 2006). Pitch, tar and resin mixes are difficult to distinguish visually and only spectrographic analysis reveals the nature of an archaeological substance containing these products (Font *et al.* 2007).

Pristine resins are crystalline and when they break they have angular or straight and sharp edges and cracks. Both pristine resins and pitches are brittle. When deposited in a smooth drop they appear glassy and transparent (Figures A.20A). In addition, resins can have a mud cracked surface, often with other plant remains embedded (Figure A.21) (Fullagar 2006). Resin is often dark in colour ranging from brown to yellow. Tar and pitch are darker than pristine resin; they usually range in colour from brown to black. Resin is birefringent and when undamaged it has a fluid and viscous feel (Figure A.20A). Starch grains, plant fibres and cells are sometimes present inside the deposit and can be observed. Resin on a tool surface or resin that was deposited for a long period can have a more granular feel.

Resins mixed with wax or other resins can lose their gloss and transparent characteristics. In general they become more gummy and sticky, and less brittle. Breaks appear more rounded and fluid than unmixed resins. Some resins used in the past were mixed with ochre, and ochre granules are visible in the resin. Mixed resins can appear more organic with globular shapes (Figure A.22 and A.23).

Plant exudate

Exudates are plant liquids, which transport nutrients, minerals and oxygen. They can contain starch grains and other cells, dissolved matter such as ions and minerals, but exudates mainly consist of water. They are much less viscous than resins. When exposed, exudates quickly dry out, leaving a stain behind. Depending on the thickness of the exudate, the stain can be a simple dark spot or a thin deposit. Often the deposit resembles a mudcracked surface with straight, but smooth edges. Sometimes starch grains are present inside the deposit. Plant exudate deposits are birefringent and semi-translucent. Exudates vary greatly in colour, from yellow to dark brown. Unlike resins,

exudates are thin layers, which are not glass-like as resin. Exudates are often only stains, whereas resins are thick patches.

Rootlets

Rootlets are sediment contaminants that accidentally adhere to artefacts and have the same characteristics as other plant tissue. They can be mistaken for use-related plant residues. Rootlets have extremely large vessels and can be confused with ancient xylem tissue. However, rootlets can have a distinct convergent cylindrical shape. Small hair can be present on the outer layer of cells. In larger rootlets, large vessels are present in the middle of the tissue and towards the outside smaller and darker cells occur.

Pollen

Flowering plants produce pollen. Pollen is comprised of microscopic grains that consist of a tough outer layer, which protects the male reproductive cell inside. Pollen grains have a characteristic shape and plant species can be recognised from them. The outer wall of pollen is named the exine and the inner wall the intine. The exine contains the resistant compound sporopollenin and this can preserve for long periods in the fossil record (Moore, Webb and Collinson 1991, Renfrew and Bahn 1996).

Pollen grains vary in size from 10 to 60 micrometre. Their shape is extremely variable and they can be, for example, round, have spikes, triangular and elliptical (Figure A.30). The grains are ever-present in the sediment and air and therefore they cannot be used in residue analysis and they must be discarded as contaminants. During this study I found many pollen grains that looked like oversized red blood cells or like round balls with spikes (Figure A.31).

2.3.2 Hyphae and spores

Most fungi are invisible to the naked eye. They live mainly in sediment, dead matter and as symbiont of plants, animals or other fungi. In ecosystems they are involved in decomposing matter and they are indispensable in nutrient recycling and exchange. Fungi can grow as thread-like filamentous microscopic structures, which are called hyphae (Figure A.32). A hypha is a long, branching cell. Hyphae are the main mode of growth and an assemblage of intertwined and interconnected hyphae is called a

mycelium (Figures A.33-A.35). The mycelium can form fruiting bodies, such as mushrooms or mould (Deacon 1997). Hyphae have chitin, a cell wall, which is build from proteins and polysaccharides. In most fungi, a membrane or the septum, divides hyphae into compartments, creating separate cells. Hyphae grow from their tips and as they grow new septa are formed behind the tip. Septa are porous to allow the transport of nutrients and cell organelles. Hyphae can also breakdown their walls in small areas and fuse together. In this way, nutrients are transported rapidly throughout a colony, even to parts where the environment is depleted (Deacon 1997).

The fruiting bodies are spore-containing structures and can be divided into: ascocarp and basidiocarp. An ascocarp consists of very tightly interwoven hyphae and can contain millions of asci, each of which contains typically eight ascospores (Figure A.33). Basidiocarp is a multi-cellular structure, which includes the spore-producing hymenium. Additionally, in asexual reproduction, spores are formed in conidia. Conidia are generally formed on the ends of specialized hyphae, the "conidiophores" (Figures A.34 and A.35) (Deacon 1997).

Fungi absorb nutrients through the mycelium. First, the hyphae secrete enzymes onto the food source (substrate), which breaks polymers down into monomers. The monomers are then absorbed into the mycelium. Some fungi develop specialized structures, which penetrate into host cells for nutrient uptake. Anaerobic fungi ferment sugars to grow and in doing so create acids and ethanol (Deacon 1997).

Hypha, mycelium and spores are contaminants, which are present in the sediment, air and on artefacts. Hyphae have a gelatinous feel and the long strands appear sticky (Figure A.37). Mycelia are shapeless masses, which also have an organic, sticky appearance. Hyphae are usually clear and translucent, but can also be brown and green in colour. Mycelia are slightly darker and vary from yellow to pinkish, but also can be black (Figure A.38). Hyphae and mycelia can be birefringent (Figure A.32). Spores are extremely variable in size, shape, surface properties and motion modes (Deacon 1997). The types, which I predominantly encountered, are black in colour with an almond shape and an elongated depression along the middle line (Figures A.39 and A.40). The other type is white and round (Figures A.41 and A.42); both are around 0.5-1 micrometre in size. The white round types can be confused with starch grains (Haslam 2006b). However, starch grains are generally bigger (>1 micrometre) and have extinction crosses. Although fungi produce spores,

they are not always associated with them and can be found in small clusters on their own.

2.3.3 Animal residues

Tissue is a group of similar cells, which have a common function. A sticky cell coating, glycocalyx, can hold tissue together or the cells can be woven together in a structure of extra-cellular fibres (Campbell 1990). There are four types of animal tissue: epithelial tissue, connective tissue, muscle tissue and nervous tissue. Animal cells are less rigid than plant or fungus cells. They have no cell wall and only contain small vacuoles, which are not clearly visible with a light microscope. In addition, animal cells do not have plastids, centrioles or peroxisomes. These cell organelles fulfil a variety of functions, from storing starch by amyloplasts and converting solar energy by photosynthesis by chloroplasts to assist in the metabolism of fatty acids and store pigments (Campbell 1990, Moran and Rowley 2008).

Epithelial tissue

Epithelial tissue is the protecting outer layer of cells in cavities of the body and it lines organs and the outside of the body. The cells are closely joined and tightly packed together in sheets (Figure A.43). The cells vary in shape from cuboidal to columnar and squamous. Some epithelia are specialised in secreting mucous or absorbing nutrients and their shape will differ (Campbell 1990, Moran and Rowley 2008).

Connective tissues

Connective tissues bind and support other tissues. Connective tissue consists of an extracellular matrix with scattered cells (Figure A.44). The nonliving matrix is a web of fibres rooted in a homogeneous matter that is liquid, jellylike or solid. There are two types of connective tissue: loose connective tissue and fibrous connective tissue.

Loose connective tissue, for example, attaches epithelia to underlying tissues (Figure A.45). The fibres in this tissue have a loose weave. There are three types of connective fibres, which are all made from proteins. Collagenous fibres are made from collagen proteins. A fibre consists of bundles of macrofibrils, which are in turn a bundle of many microfibrils. Collagenous fibres have great tensile strength. Elastic fibres are made from elastin proteins and are elastic. They resist stretching and add

resilience to connective tissue. Reticular fibres are branched fibres, which form a tightly woven fabric that joins connective tissue to adjacent tissues. Adipose tissue is specialised loose connective tissue, which stores fat in adipose cells, the adipocytes and facilitates insulation. This tissue contains many small blood vessels and is present around organs, under the skin and in specific adipose deposit throughout a body. Fat droplets are stores in adipocytes. The cells have no rigid organisation (Campbell 1990, Moran and Rowley 2008).

Fibrous connective tissue is dense because it contains many collagenous fibres. The fibres are organised in parallel bundles, maximising tensile strength. This type of tissue is present in tendons and ligaments and facilitate adherence between bones and muscle tissue. Cartilage is a specialised type of fibrous connective tissue. The many collagenous fibres are imbedded in a rubbery matrix named chondrin; this is a protein-carbohydrate complex (Campbell 1990). As a result cartilage is a strong and flexible support material. Bone is mineralised connective tissue (Figure A.45). Osteocyte cells deposit a matrix of collagen, the osteoid, and secrete calcium salts, which crystallises the matrix into hydroxyapatite. Bone is organised in repeated Haversian systems. A system is comprised of a central canal with blood vessels and nerves that is surrounded by concentric layers of mineralised matrix, the lamellae. The space between two lamellae is named the lacunae and osteocytes reside here. Osteocytes are connected by extension of the cells, the canaliculi.

Blood can be described as a connective tissue or perhaps an exudate. It consists of a liquid matrix, the plasma, in which cells and nutrients and oxygen are transported. Red blood cells are about 6-8 micrometre and white blood cells are about 7-17 micrometre (Campbell 1990, Moran and Rowley 2008).

Muscle tissue

Muscle tissue is composed of long cells, which are capable of contraction (Figure A.46). There are three types of muscle tissue, namely skeletal muscle tissue, cardiac muscle tissue and visceral muscle tissue.

Skeletal tissue is also named striated muscle tissue (Figure A.47). Muscle cells or fibres have their own plasma membrane. Fibres are subdivided into many longitudinally oriented units named myofibrils, which are again divided into sarcomeres (Figure A.46). This alignment forms dark and light bands and gives skeletal muscle tissue a striped appearance (Figure A.47). The fibres are extremely

large cells with many nuclei. They can be up to 30 centimetres long and 100 micrometre wide (Campbell 1990, Moran and Rowley 2008).

Cardiac muscle tissue is striated like skeletal tissue, but the ends of the cells are branched. The branches of one cardiac fibre attach to the branches of neighbouring fibres by highly specialized intercellular junctions, named intercalated disks. The cardiac fibres only contain 1-2 nuclei and are considerably smaller than skeletal fibres; they are about 100-micrometre long and 15 micrometre wide. Cardiac muscle tissue is present in the heart (Campbell 1990, Moran and Rowley 2008).

Visceral or smooth muscle tissue does not have striations as skeletal and cardiac tissue. Visceral cells are spindle shaped and can be much smaller than the other muscle cells. They are 20-200 micrometre long and about 5-10 micrometre wide. They only have one nucleus. Smooth tissue is present in the walls of the digestive track, bladder, arteries and other internal organs (Campbell 1990, Moran and Rowley 2008).

Nervous tissue

Nervous tissue facilitates communication between body parts (Figure A.48). It consists mainly of neurons. Neurons have a cell body with a nucleus and two or more appendages, which can receive stimuli, the dendrites or transmit stimuli, the axons. The cell body is usually between 10-25 micrometre in diameter. Depending on the animal, the appendages can be meters long, but are only about 1 micrometre thick. Glial cells or neuroglia support neuron cells; they for example, provide nutrients and propagate neuron impulses. Neuroglias form only a small part of the nervous tissue and vary in size and shape (Campbell 1990, Moran and Rowley 2008).

Muscle tissue, bone and connective fibres under the microscope

Skeletal muscle tissue and fibrous connective tissue are most abundant in animals and thus on artefacts. Muscle tissue can be birefringent, but not always (Figures A.49 and A.51). The striated tissue is characteristic (Figures A.48-A.51). In longitudinal section the parallel elongated fibres and microfibrils are visible and in cross section the cells appear circular. Muscle tissue can appear like gelatinous strands or threads (Figure A.52). The tissue has an organic and flowing feel, unlike plant tissue, which is rigid in appearance (Figure A.53). Holes can be present in the tissue. Muscle tissue varies in colour between clear to yellow and to dark orange/brown. Muscle tissue is not as

translucent as plant tissue. The ends of fibres can appear like unravelled ropes or twisted (Figures A.54 and A.55).

When muscle tissue dries out, it shrinks and discolours and in some cases it becomes very stiff. It does not resemble fresh tissue anymore and this is probably also the case when it is analysed under the microscope. When tissue shrinks, it will lose its round, flowing shapes. It cracks and shatters and it can even resemble woody fibres. Fresh muscle tissue has a very light yellow to pinkish colour, but drying will turn it into a much darker brown or orange. When rehydrated it can take its old form again, although the change in colour is permanent (Zimmerman 2001). Rehydrated mummified tissue has been successfully analysed for palaeopathological purposes and is classified to particular organs (Zimmerman 2001). However, in residue analyses tissue is not rehydrated and identification is solely based on the observation of the surface. Because exact identification of the tissues on artefacts is usually not possible, I use the general term muscle tissue in this study. In most cases muscle tissue is skeletal muscle tissue.

The fibrous connective tissues, cartilage and bone, can be difficult to distinguish from each other on artefacts. Bone is more flaky and rigid than cartilage, which is more organic or flowing, and fibrous than bone. Because collagen is a component of connective tissue, the term should not be used to indicate this tissue. In this study I refer to bone and fatty bone for amorphous connective tissue deposits and to connective fibres for collagenous, elastic and reticular fibres. Loose connective tissue can have a mixed appearance of fat, bone and muscle tissue. Depending on the relative abundance of these components, loose connective tissue is identified as fat, fatty bone, bone, connective fibres, fatty muscle tissue or muscle tissue. When I refer to connective tissue I indicate the entire category.

Bone and fatty bone are often amorphous and opaque deposits (Figure A.56). They have a greasy appearance with no characterising structure. However, angular bone flakes also occur. The flakes appear rigid and are also greasy and opaque (Figure A.57 and A.58). Bone and connective fibres are not birefringent. The fibres can have twisted or unravelled ends and also have a greasy and opaque appearance (Figure A.59). Bone deposits are usually white, but can also be light yellow; when extremely fatty, they can appear bluish in cross-polarised light (Figure A.58).

On the Sibudu artefacts I found yellow, fatty bone deposits (Figure A.60). The deposits are flaky, but also amorphous. No connective fibres are present. The deposits are opaque and have a greasy or fatty feel. The deep yellow and sometimes orange colour is striking and unlike connective tissue I encountered before. The deposits are not fat because they have too much structure. I suspect that these deposits are burnt fatty bone. The bone either burnt on the tool or it adhered to the artefact during sieving or perhaps trampling. Sibudu contains many hearths and burnt bone pieces. In the following chapter I refer to this deposit as burnt bone, but this topic needs further investigation.

Fat

Fat is stored in droplets in adipose cells and they have no rigid organisation (Figure A.61). The cells have distinct globular shapes, but when crushed or smeared, this tissue becomes amorphous (Figures A.62 and A.63). Fat from adipose tissue is white or clear and sometimes yellowish. Because bone marrow contains red blood cells, this fat is pink to red in colour. Red blood cells are sometimes present. Fat and marrow can appear opaque and sometimes bluish in (cross-polarised) light (Figure A.64). According to Fullagar (Fullagar 2006) the blue haze is caused by the secondary mineral vivianite.

Handling an artefact can leave finger grease prints on the surface (Figure A.65). Finger grease is a fatty and somewhat reflective deposit. It appears as a clear viscous layer, usually rounded or concentric. No fat granules are present and sometimes a part of a fingerprint is visible.

Blood

Blood is a special type of connective tissue or perhaps exudate (Figures A.66-A.68). When exposed, exudates quickly dry out, leaving a stain behind. Blood is reasonably thick; meaning that it contains many cells, minerals and nutrients. When dry, thick deposits generally show a mudcracked surface with straight, but smooth edges. Sometimes cells are present inside the deposit (Figure A.69). However, red blood cells are extremely small and usually only visible at magnification of 500 and 800 times. White blood cells can be larger, but are less abundant in blood. Blood is semi-translucent, but in different lights it can be relatively reflective. The colour ranges from yellow for thin blood films to black for thick films.

Hair

Hairs are formed in the deeper layers of the dermis and they are made from long keratin proteins. Hairs are cylindrical and consist of three layers (Figure 2.6). The outer layer is the cuticula and it displays cuticular scales (Figure A.70). Moving inwards, the cortex is the next layer in the hair. This layer contains amorphous dry cells and pigment granules, which are not clear under the microscope. The centre of the hair is the medulla and this also consists of tightly packed and dry cells (Figure A.71). However, the medulla is very obvious and often it is so dark that the cell structure is not visible (Teerink 1991, Bonnichsen *et al.* 2001). The scales and medulla have particular patterns that can be used to identity mammalian species (*e.g.* Teerink 1991).

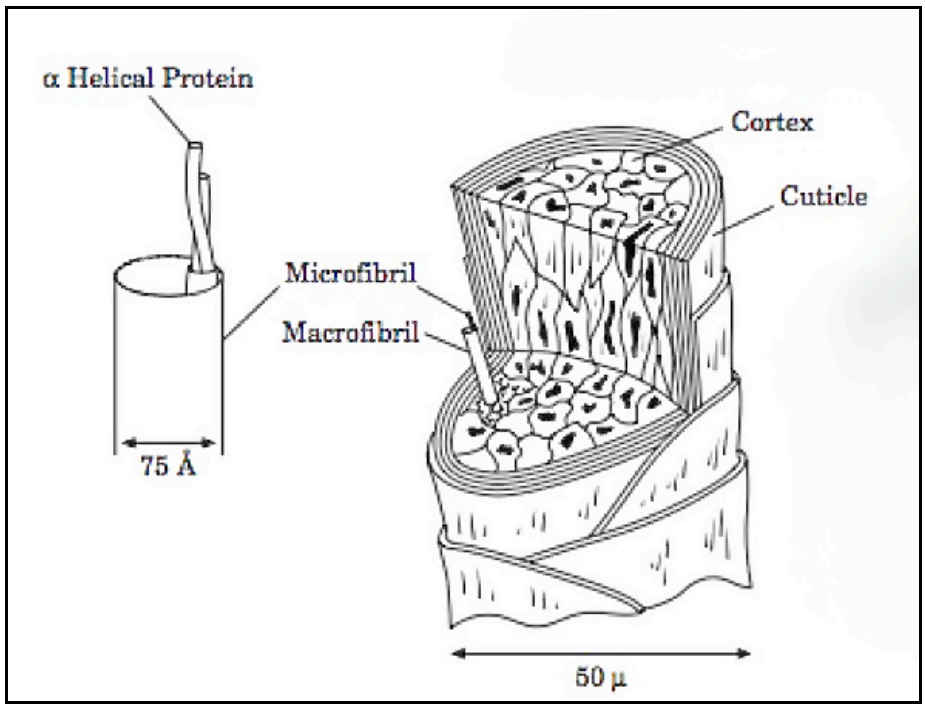


Figure 2.6: Diagram of a human hair (adapted from Bonnichsen *et al.* 2001, Figure 2, page 777).

Insect remains

In sediments, and thus on artefacts, insects or insect parts are present. The body parts are often recognisable as such and sometimes a complete insect is present (Figure A.72). Legs and torso can have small hairs. Wings have large translucent cells. Insect remains are reflective and birefringent. These remains are clear contaminants.

2.3.4 Inorganic residues

Plastic

Small square and triangular flakes of white and sometimes colourful plastic can be found in sediments and on artefacts (Figure A.73). These obvious contaminants appear similar to bone flakes. However, they are often translucent and not greasy or fatty. The flakes on the tools could potentially come from the plastic bags that the samples are housed in.

Ochre

Ochre is a red iron oxide (Fe^2O^3) or yellow hydroxide (FeOOH) (Figures A.74 and A.75A and A.75B). The iron oxide gives the mineral a red to red/brown colour, while the hydroxide gives this colloid a yellow colour. The deposit is granular. Heating dehydrates the hydroxide, turning it into red ochre. Polarised light enhances the colour and in cross-polarised light ochre is dull. Ochre is sometimes mixed with other materials, like fat and resin (Figure A.23).

Ash

Ash is a general term for light coloured and small sediment particles, but sometimes these deposits are redder or yellow (Figures A.76 and A.77). It is not possible to verify if all these sediment remains are truly ash. Sibudu sediments in particular contained a lot of ash (Figures A.78 and A.79). These deposits can appear as smears and have a greasy feel. Sometimes the small granules are individually visible and then this deposit can be confused for starch. However, starch grains are circular and ash particles are more angular. Use-related starch deposits have larger grains than ash deposits. Ash particles are often only visible at magnification 200 or higher. Small starch grains can be distinguished at magnifications of 100-200. Ash is opaque and not birefringent.

Black deposits or manganese stains

Manganese deposits are black stains, which can be slightly granular (Figures A.80 and A.81). They were identified by Dr Tom Loy and are only present on the Sterkfontein archaeological sample. The deposit can be reflective, but is not birefringent. Again the

term probably lumps different inorganic deposits together, but at this stage I cannot distinguish black sediment deposits from each other.

Nail varnish

Artefacts have numbered labels on them and these labels are often sealed with protecting nail varnish (Figures A.82 and A.83). Sometimes nail varnish is accidentally spoiled on other areas of the artefact surface. Nail varnish is clear, translucent and reflective, but not birefringent. It can have viscous strands with droplets. Air bubbles are sometimes caught in the varnish (Figure A.82). Varnish layers can be distinguished from use-related residues because they are often deposited over sediment, which adheres to the tool (Figure A.83).

Graphite and ink

Handling and deliberate marking can leave pencil and ink marks on a surface (Figures A.84 and A.85). Graphite marking covers the higher parts of the surface, leaving lower pits unmarked. Ink can be colourful, for example red or pink. Because ink is liquid it colours high and low parts of a surface area. Because these contaminants are modern, their colours are usually fresh, unlike for example, faded manganese stains. On a macro level, writing can be visible (Figure C.93).

Unknown fat-like deposit

On the experimental blanks deposited at Sibudu and Sterkfontein, I found a contaminant that is similar to fat (Figures A.86 and A.87). This residue, like fat, can appear blue in cross-polarised light. In some cases the deposit is granular under high magnifications, but this is not always so. The granular fat-like deposit could perhaps be degraded plant tissue as mentioned above. The more homogeneous type has an unknown origin.

Inclusions, breaks and wear

Usage can cause damage (micro-wear) to a tool. Micro-wear in the form of micro-fracturing, polish and striations is present on many tools in this study (Figures A.88 and A.89). Micro-wear analysis is an acknowledged method, which allows the reconstruction of tool movement and usage and processed materials. Because I am not explicitly trained in this method I cannot always determine what a particular wear

pattern on a tool represents. I have studied enough experimental tools to draw general conclusions, for example when a tool shows signs of heavy-duty processing, but I do not attempt to apply micro-wear analysis in this study. I noted visible use-wear, but did not conduct a full analysis of it. In this study "broken" means a part snapped off the tool. "Worn" or "damaged" means that use-wear is present.

Breaks can mimic bone residues. When damaged, the flint used at Zelhem and Wilhelmina Polder and the chert used at Sterkfontein appeared extremely similar to fatty bone residues (Figures A.90 and A.91). When the upper part of the surface tears, liquids can get under the tear and stain the stone. Consequently, the stained break mimics a dried liquid or thin deposit.

There is one particular break that requires consideration (Figures D.47, D.48 and D.49). During the analysis of an archaeological tool from Sibudu, SBL10 (see Chapter Six), I thought that I had found a blood residue. The characteristic mud cracking appeared to lie under the sediment deposit, typical for a use-related residue, and in (cross) polarised light the residue behaved similarly to blood. I had not observed this large residue during my primary analysis; therefore I was cautious and examined it in close detail. I noticed that the edges had no clear beginning; they seemed to disappear into the stone. As a test I pressed my finger on the sediment-covered tool's surface and when examined under the microscope the cracked surface was similar to the "blood residue". Because there is sediment on the tool, it appeared to overlie the break, implying that the breaks are use-related. I handle the samples with great care, but I created a cracked surface like a "blood residue" by accident when I pressed the tool onto the microscope stand.

Heterogenous rocks have mineral inclusions (Figures A.92-A.96). Sometimes these inclusions obscure use-related residues and confuse the analyst. Mineral inclusions are usually reflective. Sometimes a stone tool has a colour differentiation and, for example, one part of the tool is red and the other white. Analysts need to study rocks to familiarise themselves with their properties.

3 THE PRESERVATION OF MICRO-REMAINS AND IMPLICATIONS FOR PREDICTIONS

In this chapter I review the research on preservation of archaeological remains. I discuss how organic materials decay and how and under which circumstances the most common residues can preserve. In the second half of this chapter I discuss the environments at the archaeological sites and experimental locations. This leads to a prediction regarding the preservation of the experimental and archaeological residues at Sibudu, Sterkfontein, Wilhelmina Polder and Zelhem.

3.1 BURIAL ENVIRONMENTS AND WEATHERING PROCESSES

The view that one has of the past is based on archaeological finds. However, the majority of prehistoric remains are not preserved, in particular organic materials such as wood and textile. Only under exceptional circumstances, such as frozen, desiccated and waterlogged anoxic conditions, is preservation good and is there a complete picture of the past. Degradation can be described as working towards equilibrium. When artefacts and materials are deposited and buried they interact and change chemically and physically to come into balance with their burial environment (Raiswell 2001). In this section I discuss preservation and decay mechanisms. I do not discuss soil types in detail. A particular soil sets specific variables for decay, but the variables and mechanisms are what drive these processes. Soil and residue are both a target of these processes. A study of the processes and variables is more directed to answer questions concerning preservation, but the mechnisms can be applied widely. Consequently, detailed predictions about decay per soil type can possibly be made in the future. In the following section that discusses residue types and their preservation I give examples of soils, which allow preservation, from Goldberg and Macphail (2006).

Cronyn (1990) asks seven archaeological questions on why an understanding of the deterioration process of organic materials is important.

1. Can one predict where organic material may still be surviving in order to protect it or to inform excavation strategy?
2. Can the absence or condition of the material tell us something of its environment prior to and immediately after burial, or is this survival/loss more to do with the nature of the original material?
3. What effect will excavation have on organic material?
4. How can damage to the material be prevented whilst it is excavated, lifted, stored, and examined?
5. Can it be prevented *in situ*?
6. How can it be preserved for long-term study and meaningful display?
7. Can close examination of this deteriorated material tell us something about its original appearance, use and manufacture?

I want to add one more point to this list.

8. Is the preservation of material differential and could there be a bias in the data?

Points 1 to 8 are also applicable to residue analysis and preservation; residues have the same chemical building blocks as macro-remains. Microbial decay of organic materials depends on the presence of water and oxygen. In extreme dry environments such as pyramid tombs in Egypt, wooden artefacts are well-preserved. Woodworking tools from similar environments will also contain wood residues. However, a tool often contains several types of residues and this can complicate answers to the questions asked above.

For example, different materials can have distinct requirements for conservation. When materials are excavated they have to create a new equilibrium with their new dry and oxygen rich environment. This means that excavated fragile organic material usually rapidly degrades, but it can be conserved as with, for example, bog bodies and Viking ships (Caple 2001). When tools are excavated, residues are also exposed to a new environment. Lombard (pers. comm. 2007) noted that some Sibudu tools show fungi growth on tools after unearthing. I made the same observation on the experimental tools and residues from Sibudu (Chapter Four). Since tools often contain different residue types and because these cannot be conserved individually, I advise storing promising tools under dry and cold conditions, as in

well-sealed containers in a refrigerator. The cold conditions and the lack of water will considerably slow down microbial decay.

With residues one needs to be careful not to misinterpret the data bias for absence of evidence. This means that because there is no wood present at a site, it does not imply that there was no wood in the past. This point is taken for granted for the analysis of macro-remains, however, it is often forgotten for micro-remains. When looking at residues on tools, it is extremely important to realize that they also are the remnants of a period of exposure and/or burial.

Degradation or weathering is usually a combination of mechanical, chemical and biological processes. Kars (2003) describes these processes as follows:

Mechanical or physical weathering

This type of decay does not alter the chemical composition, but the original object breaks, tears and cracks into smaller pieces. The physical shape often becomes unidentifiable. Frost cracking, root working and other types of bioturbation are examples of mechanical weathering. Mechanical decay increases the surface of an object and creates cracks in otherwise durable layers such as glazing on pottery. As a result biological and chemical weathering is accelerated or initiated.

Biological weathering

In soils and sediments, microbes play the central part in decomposition processes. Micro-organisms are diverse and include bacteria, fungi, archaea and protists, as well as some microscopic plants and animals. In essence they are the agents initiating chemical processes. Oxygen is generally needed for this type of weathering, but some bacteria and fungi also operate under anaerobic circumstances. Micro-organisms are also influenced by the presence of water and the acidity of the sediment. Most micro-organisms do not favour extreme alkaline or acidic conditions. Microbial decay slows down considerably at low temperature and in absence of water (*e.g.* Tibbett and Carter 2003, Wilson *et al.* 2007).

Fungi and bacteria are the primary decomposers in most if not all, terrestrial ecosystems. I found many fungi remains and spores on experimental and ancient residues and in the following paragraphs I discuss fungi and bacteria in more detail.

Fungi grow in varied habitats, including deserts and have a worldwide distribution. Although most fungi grow in terrestrial environments, several species only occur in aquatic habitats. Fungi grow in temperatures ranging from -10°C to 65°C, but the vast majority grow best between 10°C and 30°C. Although fungi can grow in extreme acidic and alkaline environments, ranging from pH 3.0-9.0, most have a broad pH optimum between 5.0 and 7.0 (Deacon 1997). Most fungi need oxygen in at least one stage in their life cycle. Few fungi, for example some aquatic species and types that grow in cow and sheep intestines, can survive in oxygen-deprived environments. In some cases oxygen is even harmful. All fungi need the physical presence of water for nutrient uptake, enzyme release and cytoplasm maintenance. However, there are a few water-stress-tolerant fungi, which for example play a role in cereal spoilage and of other stored food products (Deacon 1997). Some of the fungi that reproduce using airborne spores need light to do this. However, the microscopic fungi, which play a role in residue deterioration, do not have air-borne spores (Deacon 1997).

Bacteria are single cell organisms, which are ever-present in all environments, ranging from nuclear waste to acidic hot springs and soil. They are visible at extreme high magnifications as, for example, with a Scanning Electron Microscope (SEM). Many bacteria are able to live at high temperatures, but only a few survive at low temperatures. pH, water and light are of little influence. However, specific species have particular optima (Campbell 1990), which I will not discuss in detail here.

Bacteria can uptake carbon nutrients through carbon dioxide, organic carbon or inorganic carbon; meaning that some species are able to degrade oil and others wood. Some metabolise using light as an energy source whereas others use energy from external chemical compounds (Campbell 1990). Most bacteria are heterotrophic, meaning that they use organic compounds as carbon and energy source. Some bacteria need oxygen to breakdown and absorb nutrients, but some obtain energy through fermentation and survive in oxygen-depleted environments. Bacteria excrete enzymes to breakdown nutrients and absorb them (Campbell 1990).

The enzymes, which fungi and bacteria use to absorb nutrients, use particular ions from the environment. If these ions or cations are not available, nutrients cannot be

absorbed. Some molecules and heavy metals inhibit enzyme activity and in both cases decay halts as the organism dies or becomes dormant (Campbell 1990, Deacon 1997).

Both fungi and bacteria can excrete breakdown by-products, such as ethanol and methane. Other microbes, such as slime moulds, manipulate their environments and create a slimy matrix in which they thrive (Campbell 1990, Deacon 1997).

Bacteria and fungi cause biological degradation of archaeological remains, but sometimes also cause chemical decay and mechanical decomposition. Sand (Sand 1997) describes the following processes:

- In ceramics microbial end products can react with ceramic compounds, creating salts that grow into crystals in the ceramic pores. This leads to mechanical stress, and eventually a pot can fall apart.
- Some bacterial reactions produce acids, which can lower the pH and acidic corrosion can become problematic.
- Some micro-organisms decay organic materials by fermentation, resulting in the production of solvents. This results in increased solubility of the archaeological artefact.
- Enzymes break components into smaller soluble parts and bacterial processes can also create emulsifying compounds. Both lead to increased solubility.
- Biofilm is a complex aggregation of micro-organisms, for example on ship hauls, but also on oil stains (Santos *et al.* 2008), which excrete a protective and adhesive matrix. The film clogs pores, which can have the similar result as salt stress. It also keeps toxic or emulsifying bacterial secretions close to the material, allowing for further corrosion.
- Bacterial decay can also produce noxious compounds such as hydrogen sulphide and nitrogen oxides. This can lead to oxidising or reducing processes and corrode materials.

Chemical weathering

During deterioration chemical reactions take place, which change and dissolve part of the components of artefacts. As a result the artefacts disappear. Water plays an important role in chemical decay, particularly in hydrolysis, but it also functions as a solvent and transporter for carbon dioxide, oxygen, ions and humic acids. When transport occurs, less favourable conditions can positively change resulting in

chemical reactions. Oxygen is needed for many chemical (oxidising) reactions and it is generally harmful for preservation, for example, rust. Acidity and alkalinity are important for chemical decay. When sediment is acidic it contains more hydrogen ions (H+) and when it is alkaline it contains more oxygen hydrogen ions (OH-). These ions actively seek out negative and positive counterparts respectively. Concentrated acidic or alkaline solutions can (aggressively) react with many other materials. Acidity and alkalinity are measured in pH values from 0 to about 14; a low pH (<6.5) means acidity, a high value (>7.5) implies alkalinity and when a solution is neutral the pH is between 6.5 and 7.5 (Goldberg and Macphail 2006).

Building on to these types of weathering, deterioration must be seen as a process, which leads to new balanced situations. Artefacts degrade because they are unstable in contact with air, water or both (Raiswell 2001). To understand and explain soil processes better I now investigate the concept of equilibrium.

Thermodynamics defines stability by the energy difference between the artefact composition and the equilibrium state. When the composition and equilibrium state both contain the same energy values, degradation seemingly halts. However, the key factor in degradation is not thermodynamic stability, but the reaction rate (Raiswell 2001). Metals, for example, are far from equilibrium states and only approach equilibrium by the slow process of oxidation. Although the energy difference is high, the reaction rate is low.

The concept of equilibrium can be illustrated by the dissolution of a salt in water. The salt will dissolve until the water is saturated and equilibrium is reached. When more salt is added it will not dissolve or some of the dissolved salt particles come together again to form salt. The following chemical reaction exemplifies:

$$aA + bB \leftrightarrow cC + dD \uparrow \text{gas}$$

“a” numbers of A react with “b” numbers of B to create “c” numbers of C and “d” numbers of D. At equilibrium as much A and B changes in C and D and the other way around. In the beginning only A and B are present and the reaction is fast, but as A and B are consumed the reaction slows down (Raiswell 2001). Therefore residues should deteriorate quickly during initial deposition and later, when equilibrium is

reached the amount of residue present stabilises; a prediction also made by Barton (2007).

Although equilibrium is predicted, the process sometimes continues until one or more reactants (A, B, C and D) are finished. This is caused by the total consumption of one reactant, for example, when C or D is a gas that disappears from the reaction. For a complete review of this field see Cronyn (1990), Raiswell (2001) and Watkinson (2001). In practice this means that organic residues are vulnerable to complete decay. Microbial decomposition of organic material releases the gas carbon dioxide and thus it is impossible to reach equilibrium. The reaction continues until the entire residue is broken down.

Describing and measuring the burial environment and the diagenetic processes leads to answers to the questions asked above. However, the burial environment is complex and it changes over time; which means that what is measured now does not have to be appropriate for ancient times. Much research has focussed on chemical and physical systems of degradation, but surface chemistry and biological organisms have added more levels of variability and complexity to the analysis of the burial environment and diagenetics (Caple 2001). Even in one area with a similar environment, deterioration can be inconsistent. For example, Tollund man, a bog mummy, whose head and feet are very well preserved, has torso and hands that are poorly preserved (Van der Sanden 1996). Forensic studies of pig burials also show differential preservation (Gill-Robinson 1999, Wilson *et al.* 2007). In general, the core of the body mass or artefact is best preserved (Turner and Wiltshire 1999, Wilson *et al.* 2007). In addition, the buried artefact also influences its burial environment. For example, in a grave the microbial load increases, leading to faster decay, liquids and gases are produced and pH values change (Wilson *et al.* 2007). Similar changes must also take place on a smaller scale.

The diversity of burial environments influences the nature and degree of degradation processes. Soil redox potential[*] and pH value are variables that provide much information on the burial environment and they are used as prime variables to

[*] Redox potential is a way to express the reducing and oxidising values of a soil. It is mainly determined by the amount of oxygen available.

describe weathering. Chesworth (1992) uses these variables to define three weathering processes, all of which are related to water activity. Without getting into as many physical, chemical and biological details as Chesworth, I describe the decomposition trends below. These trends are general trends, excluding extreme circumstances with very high or low pH values. In addition, these trends are used to describe the decay of macro-remains, but I expect them to be similar for micro-remains. Chesworth distinguishes three weathering processes:

1. The acid trend

This trend is the most widespread and occurs in humid climates where the water drainage is downwards. Because the water movement supplies ample oxygen for microbial decay, organic materials are quickly degraded. The water movements also leave abundant amounts of carbon dioxide and of course water in the soils and sediments, which results in a slightly acidic situation, containing hydrogen ions. Because hydrogen ions from the sediment will replace the mineral's own ions, pottery, glass and other minerals oxidize (Chesworth 1992).

2. The alkaline trend

This trend occurs in arid regions with a water shortfall. Due to evaporation water moves upwards, leaving soluble salts in the soil and sediment and thus creating alkaline conditions. Surface oxidation of metals can be expected as well as the capsulation of artefacts with salts. Organic materials will degrade by microbial oxidation (Chesworth 1992).

3. The reduced trend

This trend occurs where there is excess of water and absence of oxygen. In waterlogged conditions anaerobic bacteria can reduce and dissolve metals. In contrast with the other two trends, the reducing trend encourages metals to be mobile. Successive oxidation of these metals creates acidic solutions and secondary precipitation of metals. Dissolution is likely to produce pitting and etching damage on artefacts. The breakdown of organic materials often requires oxygen, and therefore these materials can preserve well. Note that local conditions can be acidic, which can affect material in other ways (Chesworth 1992).

3.2 PRESERVATION OF ARCHAEOLOGICAL MATERIAL

In some cases decay is incomplete and material is preserved. What went wrong with the deterioration process? It can be a case of one missing key ingredient or a combination of circumstances. Biological weathering is caused by micro-organisms, such as bacteria, fungi and insects that nourish on organic remains. These organisms need oxygen, water, moderate temperatures and pH values in order to survive. If there is no oxygen available for these organisms, there cannot be any decay. However, some bacteria can survive under extreme circumstances and this often means that parts of the artefact or material degrade. For example, bacteria will degrade cellulose and hemicellulose in wood, but not lignin, and as a result weak wood remains that still has many of the visual properties of normal wood (Blanchette 2000). In order to stop biological decay, one of the above-mentioned variables must be absent in the environment. For example, sand mummies in Egypt preserve because water rapidly evaporated from the bodies, creating an environment that was too dry for micro-organisms to inhabit.

Chemical weathering is difficult when there is no water to transport or dilute carbon dioxide, oxygen, ions and humic acids. Without water, hydrolysis is not possible. Chemical weathering is also problematic when there are few free cations in the environment, for example when sediment is depleted of nutrients. If there is no oxygen available, there will be no oxidation and thus less weathering. A neutral pH is also favourable for preservation. Many chemical reactions take place in highly alkaline or acidic environments.

In general the following environments are advantageous to preservation:

1. Oxygen free
2. Water free
3. Extreme pH values, particularly low values for biological preservation (for example in bogs)
4. Moderate pH values for chemical preservation
5. Nutrient or cation depleted
6. Anti-bacterial; can be extremely salty (extreme pH) or in the presence of metals
7. Low to extremely low temperatures

Now I discuss the decay process and the preservation of a range of materials. There is no written work on the preservation of archaeological residues or micro-remains. The data used in this section review work from a variety of fields, including heritage management and soil chemistry for agricultural purposes. Although unlikely, it is possible that the preservation of micro-remains works differently from that of macro-remains. I agree with Haslam (2004) that micro-environments play an important role in the preservation of residues (see Chapter Four). Otherwise, the building blocks for micro- and macro-remains are alike and their decay is thus similar. In the following section, I discuss the decomposition of materials that are often analysed in residue studies.

3.2.1 Plant tissue

Systematically arranged cells with cell walls build up wood. The cell walls are composed of varying amount of cellulose, hemicellulose and lignin (Blanchette 2000). Lignin in plant tissue can be damaged by long exposure to UV light (Smit 2003c, Smit 2003a). Mechanical weathering leads to tears and breaks in the wood and eventually wood can crumble and turn into dust (Smit 2003a). Wood is also damaged in an extreme salty and acidic environment (Smit 2003a). Moist plant tissue combined with iron or copper leads to crevice corrosion (Smit 2003a). In seawater, sulphur-reducing bacteria can grow in wood. When wood is later exposed to oxygen, the sulphur oxidises, creating a low pH, which in turn allows chemical weathering. In addition, salt crystals are formed, which causes mechanical weathering (Smit 2003a). In seawater, wood is also under attack by molluscs and crustacea, for example shipworms. These micro-organisms cause rapid deterioration of wood (Smit 2003a).

For the deterioration of residues, decay by fungi and bacteria are most important. Each of these micro-organisms attack different parts of the wood tissue and operate in different environments. Table 3.1 presents the general microbes, which cause wood deterioration. It describes which parts of the plant tissue microbes attack, the decay characteristics of each micro-organism, the environment, which is optimal for decay by this organism and the decay speed (for a full review of this topic see Greaves 1969, Blanchette, Obst and Timell 1994, Clausen 1996, Blanchette 2000, Cronyn 2001, Smit 2003a). In this paragraph I focus on the aspects of plant tissue deterioration, which are important to residue analysis.

Bacteria and fungi produce enzymes, which break down woody cell walls (Deacon 1997, Blanchette 2000). The different types of decay lead to different physical, chemical and morphological changes in plant tissue. Fungi are the most common microbes to degrade wood. They are mainly active in terrestrial environments, where oxygen is available. Fungi also need some water to breakdown wood. Bacteria degrade wood in aquatic environments, in oxygen-deprived environments and in extreme dry environments (Deacon 1997, Blanchette 2000, Smit 2003c, Smit 2003a). Different types of deterioration can occur simultaneously.

Fungal deterioration is divided into three categories: white rot, brown rot and soft rot. White rot is a common and aggressive decomposer of heartwood in living trees and of woody debris in woodland ecosystems (Blanchette 2000). Some white rot fungi attack a specific part of the cell wall; others degrade lignin and cellulose simultaneously. The latter causes rapid decay. Strength and shape losses of wood are not significant until later stages of decomposition. White rot can lead to bleaching of normal wood colouration (Blanchette 2000).

Brown rot causes quick loss of wood strength in the initial stages of decay. Brown rot breaks down carbohydrates from cell walls, leaving a lignin-rich substrate. The residual wood is brown and often breaks into cubes when dry. Some of the brown rot fungi can grow rapidly over non-nutritional surfaces, searching for nutrition and water.

Soft rot erodes the secondary walls in wood. It nourishes on carbohydrates in cell walls, the middle lamella is not eroded (in contrast to white rot fungi). These fungi cause large strength losses, but because lignin is not extensively damaged the shape is not lost. Soft rot can break down wood under more extreme circumstances than white and brown rot (Blanchette 2000).

Erosion bacteria degrade the secondary wall layers, consuming cellulose and hemicellulose. What is left is a porous lignin-rich residue. Tunnelling bacteria cause cavities or tunnels within the cell walls. These bacteria consume cellulose and some lignin. Cavitation bacteria form cavities within the secondary walls. Scavenging bacteria decompose the wood residue that is left after the other bacteria types (Blanchette 2000, Smit 2003a).

Goldberg and Macphail (2006) indicate that macro botanical remains are preserved in poorly developed soils and in soils that underwent leaching of nutrients and minerals,

such as ranker soils and podzols. Botanical remains can also preserve in, but to a lesser extent than in ranker and podzol soils, brown earth soils and luvisols. These, for example grassland and forest soils, are rich in nutrients and minerals.

Because bacteria degrade wood extremely slowly, wood is best preserved in environments hostile to fungi. This can be in an extremely dry or oxygen depleted environment and in alkaline environments.

Table 3.1: Microbes that cause wood deterioration and the processes involved. Adapted from (Blanchette 2000, 192, Table 2 and Smit 2003a, 39, Table 2).

Micro-organism	Wood components utilised	Decay characteristics	Environment	PH tolerance and temperature optimum	Degradation speed
Fungi White rot	All cell wall components	Progressive erosion of all cell wall layers.	Moist, oxygen rich	pH3-7, 25-40°C	Very rapid
Brown rot	Carbohydrates, some lignin modification	Diffuse polymerisation of cellulose	Moist, oxygen rich	pH3-7, 25-30°C	Rapid
Soft rot	Carbohydrates, some lignin modification	1 Cavities form in secondary wall 2 Progressive erosion of secondary wall	Moist-water saturated/dry, oxygen rich	pH3-9, 15-45°C	Less rapid
Bacteria Erosion	Carbohydrates, maybe lignin modification	Erosion troughs leaving large quantities of residual wall material	Dry/water saturated, oxygen depleted/oxygen rich	pH3-10.4, temp?	Extremely slow
Tunnelling	Carbohydrates and some lignin	Minute tunnels in secondary wall and middle lamella	Dry/water saturated, little oxygen/oxygen rich	pH3-10.4, temp?	Very slow
Cavitation	Carbohydrates, maybe lignin modification	Cavities form in secondary wall leaving residual material	Dry/water saturated, little oxygen/oxygen rich	pH?, temp?	?
Scavengers, etc.	1 Pit membranes 2 Modified cell wall components	1 Organisms penetrate pits and degrade wood extractives 2 Bacteria scavenge altered wood components	Dry/water saturated, oxygen depleted/oxygen rich	pH?, temp?	- (wood is already degraded)

3.2.2 Starch

Starch grains and deposits are found on archaeological tools, in pottery and in (palaeo) soils (*e.g.* Piperno and Holst 1998, Therin, Fulagar and Torrence 1999, Piperno *et al.* 2000, Balme and Beck 2002, Babot and Apella 2003, Parr and Carter 2003, Van Peer

et al. 2003, Horrocks *et al.* 2004, Perry 2004, Piperno *et al.* 2004, Fullagar *et al.* 2006, Horrocks and Lawlor 2006, Barton 2007, Horrocks, Campbell and Gumbley 2007, Piperno in press). However, the preservation of starch, particularly in soils and sediments is debated (Therin, Fulagar and Torrence 1999, Piperno *et al.* 2000). Studies demonstrate that unprotected starch quickly deteriorates (Cheshire, Mundie and Shepherd 1969, Martin 1971, Adu and Oades 1978b, Adu and Oades 1978a, Lu 2003, Lu 2006). Haslam (2006a) argues that starch preservation is generally differential and only by careful study of the preservation variables one would be able to exploit the potential of starch analysis.

The crystalline structure in starch grains is stronger than glucose, but easier to breakdown than the inorganic silica skeleton of pollen and phytoliths or the cell walls in other plant tissue. Pollen preservation is often compared to starch, but besides size similarities they are quite distinct (Piperno 1988, Lillford and Morrison 1997, Gidley 2001). Starch grains are plants' nutrition storage cells and they are packed with energy (Gott *et al.* 2006). In addition they only have a primary cell wall and are not extremely difficult to breakdown, as the plant needs easy access to this energy.

However, starch has been found in a variety of environments, ranging from extremely dry to wet and in soils and sediments with moderate pH values and extremes; starch is found in sediments and on artefacts; in temperate and warm climates (*e.g.* Ungent, Pozorski and Pozorski 1981, Ungent, Pozorski and Pozorski 1986, Tuross and Dillehay 1995, Lentfer, Therin and Torrence 2002). Some of the environments, which preserve starch are intriguing, such as (sub) tropical soils and sediments because microbial activity is high. (Piperno *et al.* 2000, Torrence 2006, table 1.1). In addition, starch is found in coprolites and a portion of starch escapes degradation in the stomach and intestines (Horrocks 2006, Samuel 2006). These are so called resistant starches. The form of starch and the method of cooking before consumption determine the amount of decay. The following resistant groups are recognised (Sajilata, Singhal and Kulkarni 2006, Samuel 2006, Lehmann and Robin 2007):

1. Physically inaccessible starches, for example partly milled grains and seeds. These are the most likely types of indigested starch to be detected archaeologically (for example, Ungent, Pozorski and Pozorski 1981, Ungent, Pozorski and Pozorski 1986, Horrocks 2006).

2. (Raw) ungelatinised starch where starch grains are packed so tightly together that enzymes cannot access them, for example, in green banana and raw potato (for example, Fullagar *et al.* 2006).
3. Thermally stable retrograded starch formed during cooling of gelatinised starch. The fully hydrated grains dry into a stronger crystalline structure, for example, as found in bread, especially stale bread, or cooked and cooled potatoes. This type of resistant starch is the toughest (for example Samuel 1996).
4. Chemically modified starch due to cross-linking with chemical reagents, for example breads and cakes.

In general, highly processes or boiled foods contain less resistant starches than raw or for example steamed foods. However, the processing mentioned under point 3 and 4 above also creates resistant starches (Sajilata, Singhal and Kulkarni 2006, Samuel 2006, Lehmann and Robin 2007). Analysis of starch in the sediments at Çatal Hüyük, Turkey, demonstrates unequal preservation of starch (Van de Locht in prep.). Starch appears to preserve best in hearth areas (Van de Locht in prep.) and heating, as discussed under point 3, possibly caused this (Van de Locht in prep.).

Resistant starches are probably not only resistant to enzymes in the intestines, but are also more resilient to bacterial and fungal attacks in soil. Note that heating, processing, digestion and freezing change the characteristic appearance of starch (Babot and Apella 2003, Gott *et al.* 2006, Samuel 2006).

Enzymes secreted by bacteria, fungi and animals, for example in saliva, breakdown the polysaccharides into absorbable sugars (Warren 1996, Barton and Matthews 2006). Fungi and animals need oxygen and some water to decompose starch and only operate under moderate circumstances. This means that the environment cannot be exceptionally cold or warm, alkaline or acidic and dry or wet. Bacteria are able to degrade starch in much harsher environments, but at a much slower rate (*e.g.* Martins *et al.* 2001). Note that extreme soil acidity also physically damages grains. Because starch is a particularly nutritious source of sugars, many bacteria and fungus species are adapted to starch degradation (Haslam 2004). Only specific circumstances inhibit microbes to degrade starch in soil.

Starch is protected from microbial attack by the presence of soil aggregates, and heavy metals, particular ions and clays. In general starch is better preserved in soils with small aggregates; fungi cannot access starch absorbed in these aggregates

(Haslam 2004). Clay, in particular montmorillenite and kaolinite, partially inactivates enzymes, preventing fungi and bacteria from decomposing starch grains (Ross 1983). Clay absorbs intermediate breakdown products of starch and this protects these products from an enzymatic attack (Lynch and Cotnoir 1956). The presence of heavy metals, such as lead and copper, also inhibits enzyme activity (Joshi, Sharma and Mishra 1993, Geiger *et al.* 1998) and anions such as iodine, sulphate and phosphate slow down degradation. Goldberg and Macphail(Goldberg and Macphail 2006) indicate that macro botanical remains are preserved in nutrient deprived rank soils and podzols and to a lesser extent in nutrient rich brown earth soils and luvisols.

Haslam (2004) and Barton and Matthews(2006) suggest that protective micro-environments, as also advocated by Kelso *et al.* (Kelso 1994, Kelso *et al.* 1995, Kelso, Ritchie and Misso 2000) for pollen, are important to starch preservation. This point is related to point 1 of the list of resistant starches. Resilient plant tissue protects starch, also in the soil and sediments, and starch granules are found in resins and in desiccated and partially charred tubers (Ungent, Pozorski and Pozorski 1981, Ungent, Pozorski and Pozorski 1986, Parr 2003). Artefacts also protect starch from fungal and bacterial influences. Grindstones, on which starch has been found on many occasions, are a good example of a protective microenvironment (*e.g.* Piperno and Holst 1998, Piperno *et al.* 2000, Babot and Apella 2003, Piperno *et al.* 2004). The pitted surface of a grindstone is an excellent “trap” for starch grains. Smaller artefacts, such as stone tools, are also likely safeguards against microbes (Haslam 2004, Barton and Matthews 2006). Pollen grains, although different in consistency from starch grains, have been found under potsherds in otherwise sterile sediments (Kelso *et al.* 1995) and it is likely that residues on tools may be similarly preserved.

Most microbes perform best in an environment with a pH between 5 and 7.5, but there are a few types that can attack starch in environments with a pH of 3.5 to 10.5. Extreme pH values inhibit or slow down decay (Martins *et al.* 2001, Haslam 2004, Barton and Matthews 2006). Water and or oxygen depleted environments reduce fungal deterioration and to some extent bacterial decay (Gould 1995, Sand 1997, Haslam 2004). Microbes also do not perform well under extreme temperatures. Some bacteria, however, can be more resistant (White and Kennedy 1988).

Table 3.2: Overview of starch conserving and degrading environments and circumstances. ?= literature is unclear on this point (See text for references).

Condition	Good preservation	Poor preservation
pH	pH < 3.5-5 pH > 7.5-10	pH 5-7.5
Temperature	< 15ºC? > 75ºC?	15-75ºC
Anions - ions	Iodine, sulphate and phosphate	Chlorine and bromine
Soil/sediment type	Clay/silt soils ranker and podzols and to a lesser extend in brown earth and luvisols	Permeable soils
Soil constitution	Small soil aggregates	Large soil aggregates
Oxygen	Oxygen depleted environments, including waterlogged environments	Oxygen rich
Soil water	Water depleted environments	Moderate water
Metals	Heavy metals (lead, copper, zinc)	
General environment	Protective micro-environments	Exposure to elements like rain
Starch type	Raw and particular processed starches	Processed and cooked starches

The preservation of starch in the archaeological record should also be explained in terms of numbers. Plants contain vast amounts of starch grains, in their storage organs, but also in other parts. When dealing with plant remains, starch is a ubiquitous component. For example, starch is also abundant in cooking and storage areas at Çatal Hüyük and less in other areas (Van de Locht in prep.), because this where starch was most abundant in the past. The chance of survival of a few grains is ensured because there are simply so many grains.

The review above leads to predictions about residue preservation (see Table 3.2), but it must be noted that starch is a sensitive material and what is good for preservation in one situation may badly affect another type of decay. Haslam, for example, notes that in the micro-environment of a 600 year-old Nubian barley grain almost no starch is preserved (Bland *et al.* 1998, Haslam 2004). Considering the dry and hot circumstances and the protective micro-environment of the grain, this is unexpected.

3.2.3 Resin

In ancient times, pristine resins and gums, tars and pitches were used as mastic and adhesive (Charters *et al.* 1993, Regert 2004, Wadley 2005, Lombard and Wadley 2007, Mizzoni and Cesaro 2007), in paint pigments (Pyatt, Wilson and Barker 2005), as medicine and funerary substance (Dixon 1972, Forrest 1982, Edwards, Farwell and

Quye 1997, Van Bergen *et al.* 1997, Edwards *et al.* 2004, Van Gijn and Boon 2006,), in mummification processes (Proefke and Rinehart 1992) and in waterproofing objects like ships and containers (Proefke and Rinehart 1992, Colombini *et al.* 2003, Connan and Nissenbaum 2003, Colombini, Modugno and Ribechini 2005, Font *et al.* 2007). An issue with tar, pitch and presitine resin is that it is often not recognised in the archaeological record (Van Gijn and Boon 2006, Regert 2004).

Wood tar and pitch and pristine resin are microbicidial, meaning that they are resistant to bacteria and fungi. The resin acid compounds are natural plant protectants and wood preservatives against herbivores and other damage. Resins are also insoluble and therefore fairly durable (Mauseth 1988, Goffer 2007), though plant gums, such as *Acacia* gum, are soluble. However, mechanical weathering and microbial decay of the organic components do occur. Often resin deterioration is a combination of microbial decomposition and mechanical weathering. De Faria *et al.* (2004) found that resin tembetá or lip-plugs from Brazil obtained a degraded and in some parts disintegrated brick-red coating, similar to iron oxide, after extended time in waterlogged conditions and later in exposed dry conditions. The resin underneath this layer was in pristine condition; the core of the artefact was best preserved as predicted by Wilson *et al.* (2007) and Turner and Wiltshire (1999). It is likely that bacteria corroded the resin while in the water and that the dry conditions later cause the objects to crumble.

Bacteria nourish on particular (organic) resin components, such as plant oils, and this results in a corroded resin "skeleton", which then is more prone to mechanical weathering or becomes soluble (for a full overview, see Wagner *et al.* 1996, Sand 1997). Comparable are situations when resins like frankincense are burnt, the organic oily component disappears, leaving behind a frail inorganic matter (Mathe *et al.* 2004). When resins are mixed with other (organic) materials the mixture can become more prone to decomposition. Organic components in the mixture decay more easily causing the left over resin to disintegrate (Sand 1997, Regert 2004).

Resins also oxidise and this leads to cracks in the surface. Resins lose their volatile component and as a result they harden over time. In both cases, resins become more brittle and prone to mechanical weathering (Cronyn 1990).

Due to the inorganic and sometimes crystalline composition, pitch and pristine resin are reasonably durable. Degradation is rapid when resin is mixed with other organic materials, which is quite common. Bacteria and fungi degrade the organic components

in resin and as previously discussed fungi do this in moderate environments. Therefore resin is best preserved in hostile environments; this is mainly in dry circumstances. However, if the environment is too dry, the resins dry out and become brittle.

Some bacteria are able to degrade organic components even under extreme circumstances, including oxygen-deprived environments. Only excessive temperatures appear to prevent all bacterial decay (Volkman *et al.* 1984). However, decay by bacteria is slow. Decay is accelerated when bacteria excrete acids, solvents, toxins or emulsifiers, which remain close to the object. In, for example impermeable soils and sediments and biofilms, these secretions corrode or dissolve resin (Sand 1997).

A moderately humid environment with moderate temperatures is probably the optimal setting for preservation.

Table 3.3: Overview of resin conserving and degrading environments and circumstances. ?= literature is unclear on this point (See text for references).

	Good preservation	Poor preservation
pH	< 5.5? > 8.0?	5.5-8.0
Temperature	< 0°C >60°C	0-60°C
Soil type	?	Impermeable
Oxygen	Deprived	Rich
Soil water	Moderate	Wet and dry, unless waterlogged
Humidity	Moderate humid	Very dry
Mixing	Pure form	Mixed with organic materials

3.2.4 Protein materials

In the following paragraphs I discuss the decay of animal proteins. The preservation of protein materials is highly debated in residue analysis. As pointed out in the Introduction, early residue studies focussed on blood and DNA residues. Although a large group of critics demonstrated flaws in protein analysis and questioned the preservation, the results from those early studies are still used as examples of protein preservation. Up to the present day there is no consensus on whether proteins preserve and whether the analysis is accurate (Hedges 1976, Anderson 1980, Loy 1983, Loy 1987, Mullis and Faloona 1987, Custer, Igenfritz and Doms 1988, Gurfinkel and Franklin 1988, Loy and Wood 1989, Hyland *et al.* 1990, Yohe, Newman and Schneider 1991, Kooyman, Newman and Ceri 1992, Loy and Hardy 1992, Smith and

Wilson 1992, Manning 1994, Downs and Lowenstein 1995, Eisele *et al.* 1995, Leach and Mauldin 1995, Evershed and Tuross 1996, Fiedel 1996, Tuross, Barnes and Potts 1996, Fiedel 1997, Newman *et al.* 1997, Garling 1998, Loy 1998a, Loy 1998b, Loy and Dixon 1998, Newman *et al.* 1998, Poinar and Stankiewicz 1999, Lawson *et al.* 2000, Kooyman *et al.* 2001, Shanks *et al.* 2001, Hortolà 2002, Lombard 2004, Lombard 2006b, Reuther *et al.* 2006, Leonard *et al.* 2007).

Biomolecular studies demonstrate that proteins can preserve long periods (for overviews see Ambler and Daniel 1991, Eglinton and Logan 1991, Thomas 1993b, Jones 1998). Consequently, these overviews are used as justification for residue analysis (*e.g.* Barton, Torrence and Fullagar 1998). Although studies demonstrate that residues can be identified using molecular analysis (*e.g.* Christensen, Walter and Menu 1992, Jahren *et al.* 1997, Langejans 2007), this is only the case with large experimental residues. Molecular analysis on microscopic ancient residues is only possible when the residue is present in large quantities, as for example in pots (*e.g.* Evershed *et al.* 2001, Evershed *et al.* 2003, Langejans 2007). In addition, molecular studies often include an analysis of the reduction complexes. Over time, large molecules, such as proteins and lipids, breakdown into smaller molecules and these are named reduction complexes. Molecular studies often do not include an analysis of proteins or lipids, but rather, they include other molecules that indicate that proteins and lipids were originally present. Even though protein molecules could preserve, and although theoretically they can be identified, it is highly unlikely. In addition, Eglinton and Logan (1991) and Ambler and Daniel (1991) clearly state that proteins only preserve under special circumstances. These include moderate temperatures, dry conditions and entombment of proteins in a non-oxidative medium, for example the mineral components of bone. Even then, amino acids from proteins undergo complete racemisation in a period of about one million years. This means that DNA and immunological analysis on samples older than one million years must be treated with great caution. It is unfortunate that Eglinton and Logan (1991), Ambler and Daniel (1991) and Thomas (1993b) used archaeological results (*e.g.* Loy 1987, Loy and Wood 1989) that have since been refuted (Ascenzi *et al.* 1985, Custer, Igenfritz and Doms 1988, Gurfinkel and Franklin 1988, Smith and Wilson 1992, Manning 1994, Downs and Lowenstein 1995, Eisele *et al.* 1995, Fiedel 1996, Fiedel 1997). Recent work is less positive than earlier work about the possibilities of DNA and

immunological analysis on ancient samples (Handt *et al.* 1994, Lindahl 1997, Cooper and Poinar 2000, Hofreiter *et al.* 2001, Amory *et al.* 2006, Leonard *et al.* 2007). This leads to another consideration; protein analysis is generally a biomolecular study, whereas residue analysis is a microscopy study. Even though proteins can preserve molecularly, they are not always visually identifiable as such. It was once thought that proteins bind to the stone tool surface and therefore survive. However, experiments suggest that protein bound to a stone surface is not protected from diagenesis (Tuross, Barnes and Potts 1996, Gernaey *et al.* 2001).

3.2.4.1 Muscle tissue

Muscle tissue has no cell walls, cellulose or great quantities of connective fibres or mineral compounds to protect it from microbial or chemical attack (Tuross, Barnes and Potts 1996, Gernaey *et al.* 2001). Forensic studies demonstrate that muscle tissue often does not last more than a few years in the ground (*e.g.* Eglinton and Logan 1991, Turner and Wiltshire 1999, Carter and Tibbett 2005, Wilson *et al.* 2007). Bacteria, aerobe and anaerobe species, and fungi degrade soft tissue. Fungi rapidly deteriorate tissue, whereas bacteria operate more slowly. Bacteria, however, can function under harsher circumstances than fungi; for example lower temperatures, extreme pH values and oxygen deprived circumstances (Wilson *et al.* 2007). Biomolecular analysis of pottery containing protein-rich foodstuff demonstrates that extremely little protein survives over time (Evershed and Tuross 1996, Gernaey *et al.* 2001). Surprisingly abundant amounts of protein are found on stone tools, which lack the protective environments that ceramics provide (Evershed and Tuross 1996, Gernaey *et al.* 2001, Lombard 2006b). The experiments suggest that protein bound to a stone surface is not protected from diagenesis (Tuross, Barnes and Potts 1996, Gernaey *et al.* 2001). Biomolecular studies intimate that proteins decay rapidly in warm climates, at open-air sites, particularly when there is a water flow through the site and when microbial activity in soil/sediment is high. In addition sunlight also destroys proteins and protein parts (Gernaey *et al.* 2001).

Experimental forensic studies on buried pigs present the following chain of events for putrefaction of soft tissues in particular: the first sign of decay is blue grey blotches, which appear on the skin. Next, hairs come loose from the skin. The skin and soil interface turn grey, and by then, the skin epidermis is easily rubbed off. The

underlying tissues, which are now partly exposed, dry out and turn black. Tissues become soft and mousse-like. The fat layers are readily separated from the underlying unexposed pink muscle tissue. By now the skin is black and easily detached; it is difficult to separate the skin from the surrounding soil. Next the skin and soil interface becomes indistinct and only small amounts of intact and coherent muscle tissue are present in the deeper part of the burial. Finally the tissues liquefy and are absorbed in the soil. Flies lay eggs in the tissue, but only when it is accessible and exposed. Fly larvae feed on the decomposing tissue and accelerate decay. Other organisms, such as earthworms also feed on the soft tissues, but their influence depends on the burial depth and the bioactivity in the soil (Turner and Wiltshire 1999). The 150-day experiment described above was conducted in a heavy clay soil in Hertfordshire, UK. The decomposition rate varies between soils, locations and climates (Hopkins, Wiltshire and Turner 2000, Wilson *et al.* 2007). Unfortunately none of the above used forensic studies describes in detail how soft tissue changes in appearance. Turner and Wiltshire (Turner and Wiltshire 1999) state that when muscle tissue is exposed, it dries out and turns black and, when buried, it is pink and becomes mousse-like. The latter probably indicates loss of cell structure.

Dry, preserved soft tissue, human and animal, is mummified. The durability of skin and tissue depends on the processing method, namely tanning, tawing and chamoising. Processing can be done artificially to create leather or to preserve a body or it can take place naturally. The following types are recognised for leather: vegetable tanning, alum tanning and mineral (chrome) tanning and aldehyde tanning with smoke (Dirksen 1997, Smit 2003b). Purposeful mummification often depends on the extraction of water and on creating an antibacterial environment, as for example the natron bath of Egyptian mummies. The removal of weak non-muscle tissues, such as intestines and brains, also lead to better preservation; particularly because these tissues are a source of bacteria and decay (Evershed and Connolly 1994, Van der Sanden 1996). Natural tanning follows the same lines as artificial tanning. It can occur under extremely dry circumstances, for example mummification in hot and arid regions, and dry freezing, and in environments that contain tanning ingredients, like peat and solid oak tree trunk coffins (Painter 1991, Painter 1998, Painter 2003, Smit 2003b, Goldberg and Macphail 2006). Prehistoric leather, skin and soft tissue is preserved in waterlogged and anaerobic environments, which usually have obtained tanning properties from sphagnan moss or humid acids in peat bogs; in combination

with salt and metals (salt and metals, like copper, have anti-bacterial properties); and in extreme dry and cold environments (*e.g.* Höpfel, Platzer and Spindler 1992, van Klinken and Hedges 1995, Groenman-van Waateringe, Kilian and Van London 1999, Smit 2003b).

Muscle tissue and skin only preserve under extremely extraordinary circumstances by natural tanning. Natural tanning, in bogs and from drying and dry freezing, leads to leather-like tissue. In addition, dark environments preserve proteins better than ones exposed to sunlight. Muscle tissue shrinks and discolours and in some cases it becomes very stiff. It does not resemble fresh tissue anymore and this is probably also the case when analysed under the microscope. When rehydrated it can take its old form again, although the change in colour is permanent (Zimmerman 2001). Rehydrated mummified tissue has been successfully analysed for palaeopathological purposes and classified to particular organs (Zimmerman 2001). However, in residue analyses, tissue is not rehydrated and identification is solely based on the observation of the surface.

Table 3.4: Conditions for muscle tissue preservation and decay. ? = literature is unclear on this point.

	Good preservation	Poor preservation
pH	With salts >9.0 or acidic when waterlogged	<9.0
Temperature	<10°C or >30°C	Moderate
Soil type	Peat	Permeable with water flow
Oxygen	Oxygen deprived in waterlogged circumstances in bogs	Oxygen rich
Soil water	Rapid desiccation (also freeze dry) Waterlogged circumstances in bogs	Moist and fluctuating circumstances
Humidity	Extremely dry atmosphere	Humid atmosphere
UV	In darkness	Exposure to sunlight

3.2.4.2 Blood

Blood, like muscle tissue, consists mainly of proteins. Biomolecular analysis demonstrates that extremely few proteins can survive long time periods (Evershed and Tuross 1996, Gernaey *et al.* 2001). However, surprisingly abundant amounts of protein are found on stone tools (*e.g.* Loy and Wood 1989, Loy and Dixon 1998, Williamson 2000b, Kooyman *et al.* 2001, Shanks *et al.* 2001, Lombard 2004, Williamson 2004, Lombard 2006b). Although it was once thought that proteins bind to the stone surface and therefore survive, experiments suggest that protein bound to a stone surface is not protected from diagenesis (Tuross, Barnes and Potts 1996,

Gernaey *et al.* 2001). Biomolecular studies suggest that proteins decay rapidly in warm climates, at open-air sites, particularly when there is a water flow through the site and when microbial activity in soil/sediment is high. In addition sunlight also destroys proteins and protein parts (Gernaey *et al.* 2001).

Ancient blood is cracked and the deposits are torn and discoloured. Therefore blood is usually chemically identified by the detection of proteins. Smith and Wilson (Smith and Wilson 2001) describe the problems that are involved with these kinds of chemical analyses. A major issue with blood analysis is that proteins preserve badly and in quantities that are too small for chemical analysis. This is particularly problematic for proteins on buried artefacts (Gurfinkel and Franklin 1988, Cattaneo *et al.* 1991, Tuross, Barnes and Potts 1996). Chemstrip and Hemastix tests, and also haemoglobin crystallisation, are unreliable. Other techniques, like Enzyme Linked Immunosorbent Assay (ELISA) and Radioimmunoassay (RIA) are more promising, but due to their complexity they are less used (Smith and Wilson 1992, Smith and Wilson 2001).

Hortolà (2002) describes experiments on blood preservation and visual identification of blood. Blood was smeared on three sets of tools, two sets were dried in the sun and one set away from sunlight. The dried samples were stored in a non-sterile room with moderate temperature and humidity. After 7 to 10 years the samples still preserve red blood cells, negative red blood cells replicas and red blood cells with a moon-like shape. Although sunlight influences the immunoreactivity (Tuross, Barnes and Potts 1996), moderate exposure to natural UV light does not affect red blood cells morphologically (Hortolà 2002).

Except for Hortolà (2002), all the studies described here are molecular investigations. It is unclear, if Cattaneo *et al.* (1993) and other researchers also visually observed the blood residues. However, blood appears to preserve under desiccated and frozen conditions (Smith and Wilson 2001, Hortolà 2002). In addition, Cattaneo *et al.* (1993) remark that blood residues are better preserved when dried and that cave sites and desiccating conditions are probably more suitable for preserving blood. In addition, they comment that low rainfall is also good for preservation Gurfinkel and Franklin's (1988), and examples of wet samples deteriorating quickly supports their idea. Proteins deteriorate during prolonged exposure to sunlight (Tuross, Barnes and Potts 1996).

Table 3.5: Conditions for blood preservation and decay. ? = literature is unclear on this point.

	Good preservation	Poor preservation
pH	?	?
Temperature	> 0°C	Warm
Soil type	?	?
Oxygen	?	?
Soil water	Dry-desiccated	Wet/moist
Humidity	Dry-moderate	Very humid
UV	Dark environment	Prolonged exposed to sunlight

3.2.5 Fat

The decomposition of hardy insoluble fats, oils and other lipids by hydrolysis or oxidation is called rancidification. Complex fatty acid molecule chains break down into smaller soluble fatty acids and then they oxidise. Saturated animal fats are more durable than unsaturated plant oils. Light and oxygen rich environments increase the rancidification rate.

Fat is part of many materials, including plant waxes, beeswax, animal fat, dairy fat, plant oils, natural bitumens and plant resins (Evershed *et al.* 2001). Lipids are found in many archaeological contexts, but they are extensively analysed when preserved on pottery (*e.g.* Condamin *et al.* 1976, Patrick, Koning and Smith 1985, Evershed *et al.* 2003, Copley *et al.* 2005a, Copley *et al.* 2005b, Copley *et al.* 2005c, Spangenberg, Jacomet and Schibler 2006). The unglazed pottery creates a "protective micro-environment for fats that functions as "molecular sieve" or "trap" preserving lipids during burial over many millennia" (Evershed *et al.* 2001, 332). Lipids and their reduction complexes have been found in soil and sediment deposits and used as indicators of changes in agricultural practices and soil use and the use of manure (*e.g.* Eglinton and Logan 1991, Simpson *et al.* 1998, Bull *et al.* 1999). Fat is also preserved in human and animal remains (*e.g.* Kuksis *et al.* 1978, Evershed *et al.* 1995). However, fat in these contexts is molecular and unfortunately little research has been done on visible deposits.

Forensic studies and analysis on bog bodies demonstrate that deteriorated fat is recognisable as adipocere. Under moist, warm and anaerobic circumstances and in mildly alkaline environments, bacteria decompose fat and fatty tissues into adipocere (Forbes, Dent and Stuart 2005a, Forbes, Dent and Stuart 2005b). Adipocere is a solid greyish-white substance. When bound to potassium it is wax-like and when bound to sodium it is hard and crumbly (Forbes, Dent and Stuart 2005b, Vane and Trick 2005). Over time, adipocere also becomes harder and more brittle (Forbes, Stuart and Dent

2005). It consists of saturated and unsaturated fatty acids, salts of fatty acids and hydroxy- and oxo-fatty acids (Forbes, Dent and Stuart 2005b). Adipocere is an insoluble and durable substance and can preserve for long time periods (Evershed 1992, Forbes, Stuart and Dent 2005).

Although insoluble, fat can easily mix with sediment or soak into porous materials, in which case it is no longer visible, but may become a residual stain on the stone tool remains. In addition to biogenetic decay, auto-oxidation, which is encouraged by free radicals, occurs under the influence of sunlight. Because lipids are complex molecules, they are difficult to break down and therefore these residues and their breakdown products probably last (Evershed *et al.* 2001).

Table 3.6: Conditions for fat preservation and decay. ? = literature is unclear on this point.

	Good preservation	Poor preservation
pH	Mild alkaline	?
Temperature	Moderate and freezing	?
Soil type	?	?
Oxygen	Anaerobic	Aerobic
Soil water	Waterlogged-moist	?
UV	Dark environment	Light environment

3.2.6 Bone

Bone is a resilient organic material and often the only organic matter preserved. It consists of a mineral component, named bioapatite (70%), and an organic component (30%). The organic part mainly consists of a matrix with connective fibres and other non-collagenous proteins and water (Jans 2003). Compact and large bones preserve better than porous (spongeous) and small bones (Von Endt and Ortner 1984, Willey, Galloway and Snyder 1997, Jans 2003). Burnt bone no longer contains elastic connective tissue and apatite crystals recrystallised into larger and more stable crystals. Burnt and cooked bone is resilient and less affected by microbial decomposition (Nicholson 1998, Jans 2003). However, volume changes due to recrystallisation sometimes lead to fragmentation and deformation. Because connective tissue is lost, burnt bone can become brittle and crumble easily (Stiner *et al.* 1995, Jans 2003).

Insects and roots can damage the bone surface, but the main decomposers of bone are fungi, bacteria and cyanobacteria in marine environments (Marchiafava, Bonucci and Ascenzi 1974, Hackett 1981, Bell, Boye and Jones 1991, Jackes *et al.* 2001, Jans *et al.* 2004). Fungi and bacteria secrete enzymes to break down and absorb

the organic and mineral components from bone. In doing so they create microscopic tunnels in the bone. Fungi cause Wedl or centrifugal tunnelling. The tunnels are 5-10 micrometre in diameter and they are empty, indicating absorpsion of organic and mineral parts from the bone. Bacteria create three types of tunnels: "linear longitudinal", "budded" and "lamellate". Bacterial tunnels (average diameter 0.5 micrometre) are smaller than fugal tunnels and the interior of the tunnels consists of small pores, an indication that only organic components are absorbed (Hackett 1981, Jans *et al.* 2004). Jans *et al.* (2004) suggest that the budded tunnels are a prerequisite to the lamellate formation or lamellate is a subcomponent of budded tunnelling.

Intestinal and endogenous bacteria of a corpse instigate bacterial attack on bone. This explains why bone fragments, not affected by putrification, often do not suffer from bacterial decomposition (*e.g.* Nicholson 1998). Bacteria can break down bone under more extreme circumstances than fungi. Bacteria are able to operate in, oxygen and water deprived environments and at lower/higher temperatures and pH values than fungi, but under extreme circumstances bacterial decay is ceased. Poisonous materials, like copper, lead and mercury, humic acids in waterlogged conditions, extreme temperatures, rapid desiccation in extreme dry environments or the absence of endogenous bacteria (in for example children or gutted carcasses) prevent bacterial decomposition (Janaway 1987, Mant 1987, Grupe 1995, Janaway 1996, Nicholson 1996, Bocherens *et al.* 1997, Jans *et al.* 2004, Nielsen-Marsh *et al.* 2007).

Fungi, however, are ubiquitous in oxygen rich and semi-humid environments and decompose bone when environmental conditions are favourable. Oxygen and water depleted environments inhibit fungal degradation (Child 1995), but note that extreme dry environments can also lead to deformation (Grupe 1995).

Collagenous, elastic and reticular proteins maintain the strict order within mineral apatite crystals and when it disappears bone is prone to chemical weathering (Pate, Hutton and Norrish 1989). Depolymerisation of the connective tissue proteins and hydrolyses cause chemical degradation of bone in soil (Collins *et al.* 1995, Jans 2003). Specific soils and sediments do not guarantee decay, but the following interrelated processes lead to changes in the mineral component of bone:

- Soil minerals precipitate in bone pores and bone can crumble and fragment (Pate and Hutton 1988). For example, high concentrations of calcium in the

soil matrix lead to the formation of large calcium crystals (Piepenbrink 1989, Child 1995).

- Diagenesis of bone ions can occur, leading to demineralisation of bone (Gordon and Buikstra 1981). In acidic soils, ion exchange and thus demineralisation is rapid (Gordon and Buikstra 1981). For example, low concentrations of phosphates in the soil cause demineralisation (Child 1995).
- Apatite can recrystallise and crystals can mature. Apatite is soluble at low pH values and the bruhite crystals left behind are larger, causing the bone to fragment. In addition, brushite is soluble and bone quickly disappears (Piepenbrink 1989, Child 1995, Crow 2008).

In terrestrial surroundings, two environments can be distinguished:

1. Corrosive environments, which are permeable, acidic and oxygen rich. They lead to leaching of minerals, loss of bone apatite and an increase in bacterial and fungal decomposition respectively (Kars and Kars 2002, Jans 2003). Nielsen-Marsh *et al.* (Nielsen-Marsh *et al.* 2007) demonstrate that that the difference in proportion of completely deteriorated bone more than doubles between soils with a pH 6.0 and soils with a pH 5.5. Low concentrations of phosphates in the soil also lead to demineralisation. High concentrations of calcium in the soil matrix lead to the formation of large calcium crystals, which fragment bone (Jans 2003). Soil acidity proves the most destructive decomposer, and in corrosive soils chemical breakdown is more important than microbial attack of bone. In corrosive soils, macroscopic damage is obvious because mineral dissolution rates are higher (Nielsen-Marsh *et al.* 2007). Soils, which are corrosive include: leaching rank soils and podzols. Secondary crystal growth in calcite rich rendzina and solonetz soils also damage bone (Goldberg and Macphail 2006).
2. Conserving or benign environments contain a large percentage of organic matter, are pH neutral or slightly alkaline and contain some calcium and they are oxygen or water deprived (Kars and Kars 2002, Jans 2003, Crow 2008). In addition, the microbicidal circumstances described above also ensure preservation. Extreme dry environments also preserve bone although not always in pristine condition (Grupe 1995). Bones in benign soils demonstrate more microbial attacks than bones in corrosive environments, leading to

histological damage and protein loss and a general bimolecular decay (Nielsen-Marsh *et al.* 2007). Because brown earth and luvisols are not acidic or leaching and are in addition nutrient rich, but do not contain high amounts of calcite, they can be benign soils (Goldberg and Macphail 2006).

Figure 3.2 presents a schematic overview of bone decay and preservation trajectories.

Bone can preserve in marine contexts; the organic components are well preserved by seawater, but the mineral part contains more calcium, leading to fragmentation. Algae and (sulphur reducing) bacteria decompose bone in marine contexts (Jans *et al.* 2004). Although oxygen depleted, bone does not preserve well in waterlogged conditions, such as bogs. The humic acids in bogs initiate chemical decay. Because flowing water is absent, bacteria and their by-products remain close to the bone and degradation may even be enhanced in this environment (Child 1995, Gernaey *et al.* 2001).

In most bones the mineral phase rapidly dissolves, leading to a complete loss of bone (Nielsen-Marsh *et al.* 2007). Early taphonomy is extremely important for bone preservation in benign soils and sediments. The initial state of preservation of a bone, for example whether the bone is processed or part of a cadaver, or whether it is burnt, determines preservation (Smith *et al.* 2007).

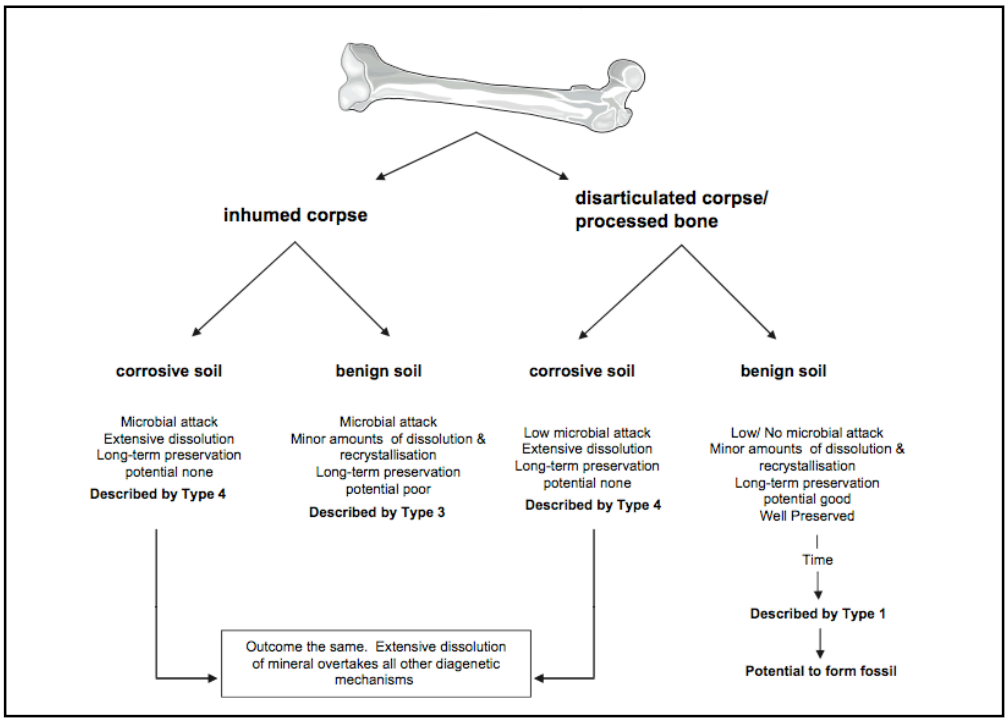


Figure 3.1: Diagenetic end-points for bones in terrestrial environments, taking into account the effects of burial environment and taphonomy (From Nielsen-Marsh *et al.* 2007, 1529). Smith *et al.* (Smith *et al.* 2007) recognise four types of preserved bone: Type 1: Is well preserved bone. It contains a high percentage protein, has a high Oxford Histology Index score, a high C/P ratio and a low Mineral Crystallinity Index and low porosity values. Type 2: Underwent accelerated collagen or protein hydrolysis. It contains a low percentage protein, a highly altered mineral index, but with a high Oxford Histology Index score, a low C/P ratio, a high Mineral Crystallinity Index and increased porosity with small pores. Type 3: Is microbially attacked bone. In this type protein tends to decrease, there is a decreased Oxford Histology Index score, occasional hyper-mineralised edges and mineral alteration, an increasing Mineral Crystallinity Index and an increase in porosity with medium pores. Type 4: Underwent catastrophic mineral dissolution. It contains a low percentage protein, has a low Oxford Histology Index score, a low C/P ratio, high Mineral Crystallinity Index and high porosity values with large pores.

Table 3.7: Overview of conditions for bone preservation and decay.

	Good preservation	Poor preservation
pH	>7.0 <9.0	<7.0
Temperature	Extremes	Moderate
Soil type	Impermeable and brown earth and luvisols	Permeable
Oxygen	Depleted	Rich
Soil water	Dry or waterlogged with few humic acids	Moist to waterlogged
Atmosphere	Dry-moderate	Humid
Pre-burial	Dismembered pieces, burnt, cooked	Cadavers, fresh bone
Other	Presence of heavy metals	-

3.2.7 Hair

Hair is found in many different archaeological settings, for example, on stone tools from southwest Germany, on frozen mummies in Siberia, on desiccated Egyptian mummies and on waterlogged bog bodies (*e.g.* Lubec *et al.* 1987, Brothwell 1996, Iacumin, Davanzo and Nikolaev 2005, Amory *et al.* 2006, Hardy, Bolus and Conard 2008). The hard cuticle outer surface and the strong disulfide bound molecule structure prevent microbial attack. Hair proteins are hydrophobic and not soluble in water. In addition, fatty acids in wool wax have an antimicrobial character and protect hair from microbes. Consequently, hair is durable (Bonnichsen *et al.* 2001, Szostak-Kotowa 2004). However, hair and wool can contain impurities, which are susceptible to a microbial attack. Deterioration is mainly caused by fungi, but also by bacteria. When hair or wool is processed, and when they are chemically, photochemically or mechanically degraded, they become more susceptible to microbial attack (Szostak-Kotowa 2004).

Degradation is often only visible when it is well advanced. Fungi first attack the cortical cells, leading to a loss in scale structure. The scales partially lift up and curl up or the cortical cells are released from the fibre. Hyphae bore holes into the now less resistant fibres. Attacked fibres exhibit fibrillation and frayed fibre ends. Fibres attacked by bacteria are reduced to hollowed out tubes; the cortical scales fall off and the fibres exhibit fibrillated ends. Hair often becomes darker in colour when it is degrading (Peacock 1996b, Peacock 1996a, Szostak-Kotowa 2004). Hair is preserved in close proximity to heavy metals, for example copper. These metals are poisonous to microbes (Janaway 1985).

High humidity, moderate temperatures and acidity advance microbial growth on hairs (Szostak-Kotowa 2004). However, according to Gochel, Belly and Knott (1992) high acidity prevents microbial decay of wool and an alkaline environment increases decay. Sibley and Jakes (Sibley and Jakes 1984) demonstrate that wool is sensitive to alkalinity and that acidic environments with few salts preserve hair best (also see Smit 2003c). Oxygen depleted, water deprived or waterlogged environments and extreme cold or warm environments halt fungal decay. Peat soils with their waterlogged properties can preserve hair (Goldberg and Macphail 2006).

Table 3.8: Overview of the conditions for hair preservation and decay. ?= no or conflicting data.

	Good preservation	Poor preservation
pH	7,5 and >5 or <8?	?
Temperature	Extremes	Moderate
Soil type	Peat	-
Oxygen	Depleted	Rich
Soil water	Dry or waterlogged	Moist
Humidity	Dry-desiccated	Humid

3.2.8 Preservation synthesis

The paragraphs above give a detailed description of the preservation of organic materials. Table 3.9 is an overview of the advantageous conditions under which materials preserve. Often not one factor determines preservation, but it is a combination of variables. For example, acidic waterlogged conditions are more favourable for preservation than alkaline waterlogged conditions. From paragraphs 3.2.1 to 3.2.7 it is apparent that sediment pH and moisture are the important factors influencing preservation. Ion saturation of the sediments is also important, for example calcite ions prevent bone decay. However, often no information is available in the literature. In general, a high ion saturated soil and soil with ion equilibrium is best for preservation of organic remains (Crow 2008). In Figures 3.2 to 3.9 I present an overview per residue type of the quality of preservation in environments with different soil moisture and pH values. Note that the figures are for moderate temperatures. From the Figures it is clear that there is not one optimum for all organic material types.

The literature is not always clear on how all remains preserve. For example, adipocere formation and molecular analysis of fatty acids is extensively discussed, but the preservation of fat as a visually identifiable micro-remain is overlooked. This means that the data in Figure 3.7 are not complete. It is possible that I over indicated “poor preservation” because there is too little information available.

Table 3.9: Overview of advantageous conditions for the preservation of particular residues. - = not advantageous; X = advantageous; N.i. = no information available; ?= conflicting information.

Residue type	Atmosphere dry	Atmosphere humid	Heavy metals	Soil type	Soil constitution	Oxygen depleted	No UV light	Temperature	Other
Plant tissue	X	-	N.i.	Ranker and podzols and to a lesser extend in brown earth and luvisols	N.i.	X	X	Extremes	
Starch	X	-	X	Ranker and podzols and to a lesser extend in brown earth and luvisols	Clay/silt sediments and small aggregates	X	N.i.	Extremes	Processed starch preserves better than raw starch and protecting micro-environments also help preservation
Resin	N.i.	N.i.	N.i.	N.i.	N.i.	X	N.i.	Moderate	In pure form
Muscle tissue	X	-	X	Peat	N.i.	Waterlogged in bogs	X	Extremes	
Blood	X	-	X	N.i.	N.i.	N.i.	X	> 0°C	
Bone	X	-	X	Brown earth and luvisols and to a lesser extend rendzina and solonetz	Impermeable	X	N.i.	Extremes	Waterlogged with few humic acids. Dismembered, burnt and cooked pieces preserve better than carcasses. Calcium in the soil protects bone from decay.
Fat	N.i.	N.i.	N.i.	N.i.	N.i.	X	X	Warm	Under warm and moist circumstances fat transforms into adipocere
Hair	X	-	?	Peat	N.i.	X	X	Extremes	



Figure 3.2: The quality of preservation of plant material in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).

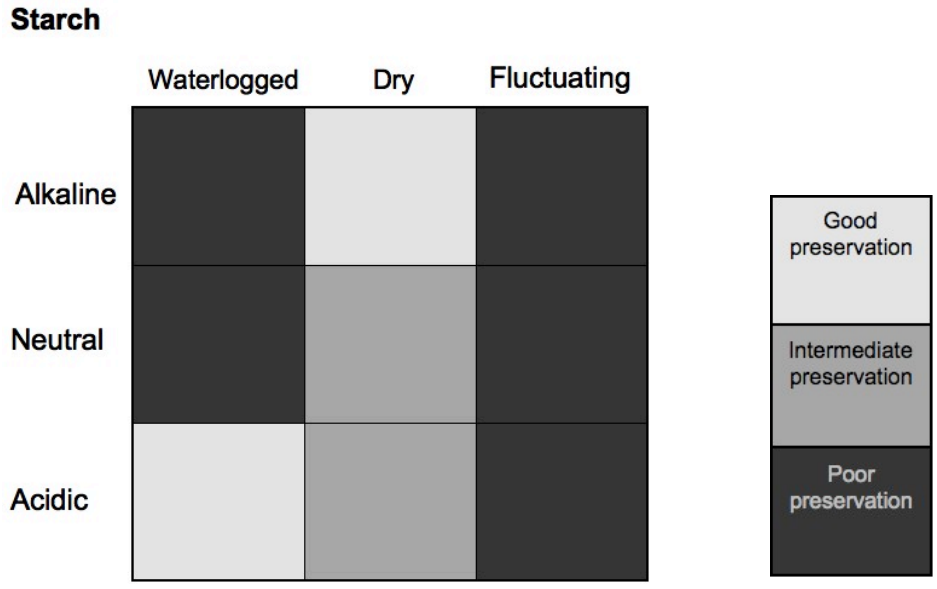


Figure 3.3: The quality of preservation of starch in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).



Figure 3.4: The quality of preservation of resin in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).

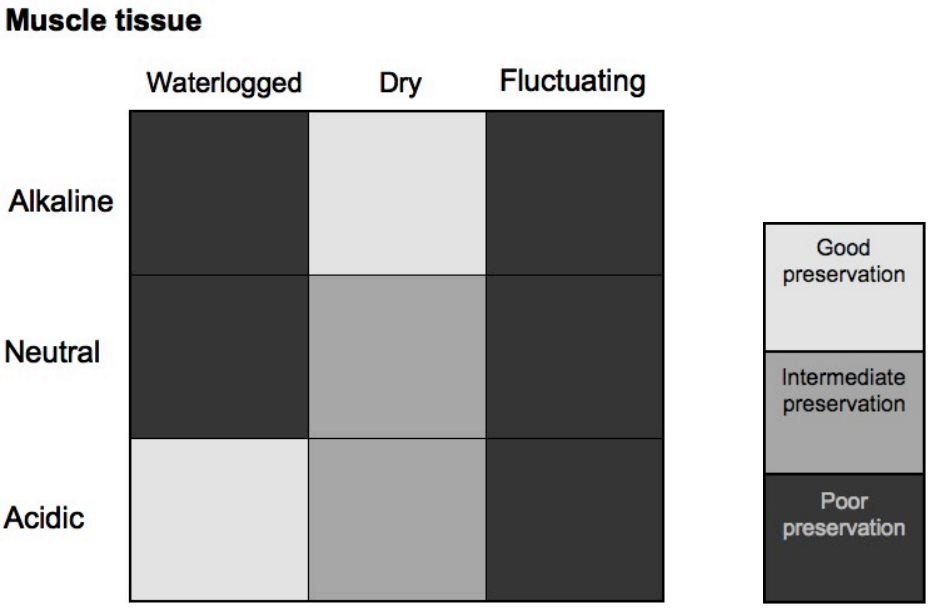


Figure 3.5: The quality of preservation of muscle tissue in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).

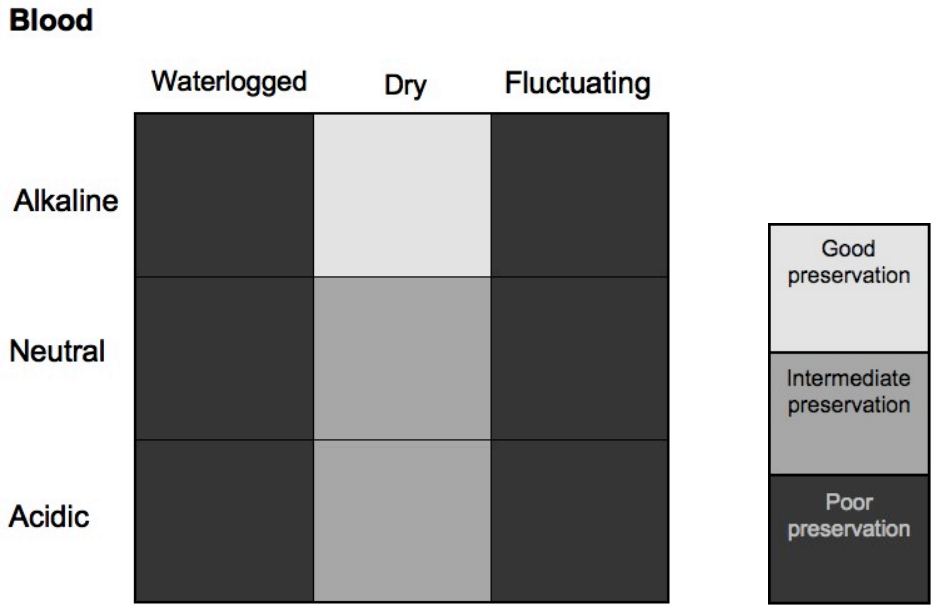


Figure 3.6: The quality of preservation of blood in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).

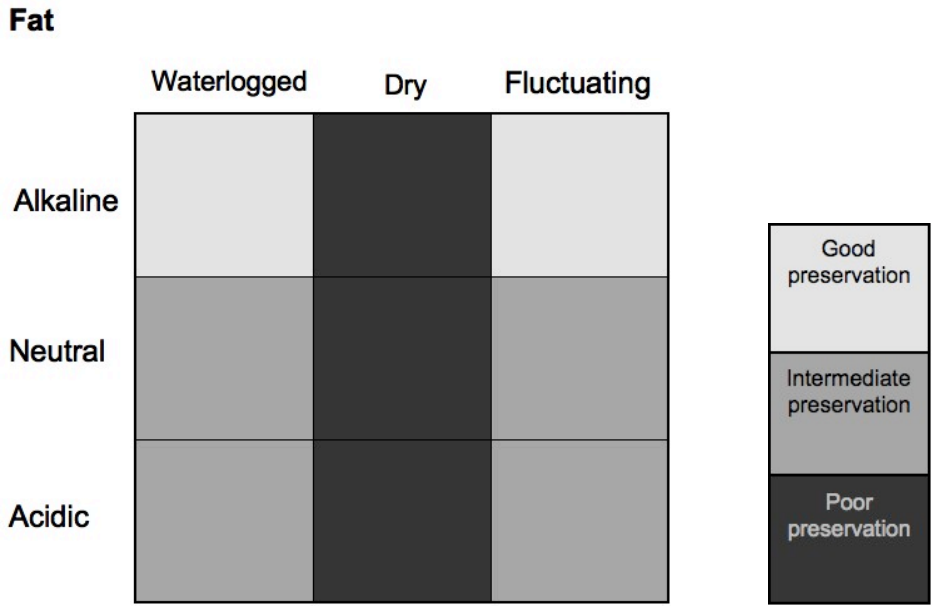


Figure 3.7: The quality of preservation of blood in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).

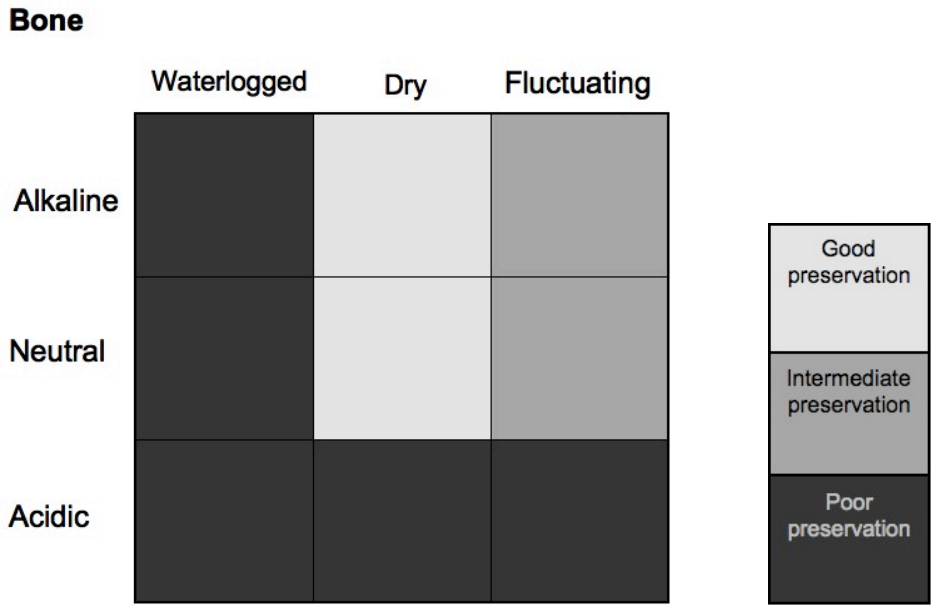


Figure 3.8: The quality of preservation of bone in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).

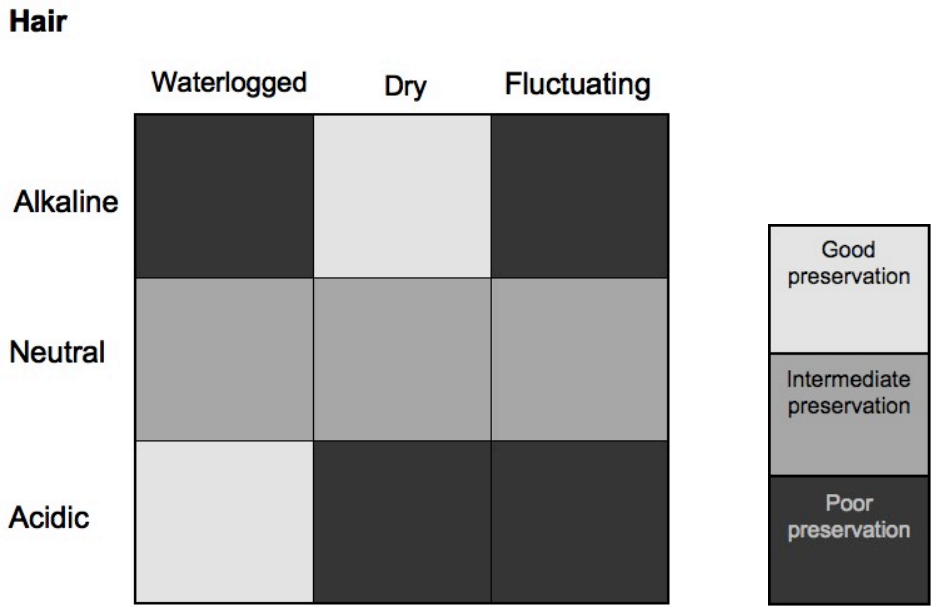


Figure 3.9: The quality of preservation of hair in a variety of environments at moderate temperatures (Based on information from the paragraphs above and from (Crow 2008, Figure 5, 2272).

3.3 DESCRIPTION OF THE ARCHAEOLOGICAL AND EXPERIMENTAL ENVIRONMENTS AND SETTINGS

In the following paragraphs I discuss the environments at the sites and I predict what residues can or cannot preserve in these environments. The environmental criteria from table 3.8 are applied to a summary of the environmental conditions at the sites in table 3.11. Soil moisture, pH and atmosphere humidity are the most important criteria. The other criteria alone cannot ensure good residue preservation. Some criteria are linked. For example, fat preserves in warm and moist circumstances, but only when the environment is anaerobic. For the archaeological samples I cannot predict definite preservation conditions because there are too many uncertainties, such as time.

3.3.1 Sterkfontein Cave

The Sterkfontein Caves are situated about 60 kilometres north of Johannesburg, Gauteng province, South Africa. The natural vegetation in the modern Blaaubank River Valley is a mixture of open grassland, bush and woodland. A wide variety of bushes and shrubs is present on the rocky hills and ridges. The site is located about 30 metres above the river and is about 550 metres away from it (Avery 2001). Due to altitude differences it is complicated to give exact mean temperatures and precipitation values for Sterkfontein. In this study I use data from the Zuurbekom weather station because the altitude is closest to Sterkfontein's. The mean winter monthly minimum temperature is -2°C and the mean summer maximum is 27°C. The mean annual precipitation is 660 millimetres of which 14% is winter rainfall (Weather-Bureau 1986, Avery 2001).

The archaeological settings

The underground cave systems are karst caverns in dolomitic limestone. Acidic groundwater dissolves parts of the karst rock along planes of weakness and caverns form while the rock is still below the water table. After a drop in the water table, stalagmites and stalagtites form through drip within the closed cave. Much later in time, starting from about 3,5 Ma ago, the slightly acidic groundwater created narrow vertical shafts, which opened these caves to surface. From then on material, including

sediment and biological remains were deposited into the caves. Once in the cave the material accumulates in a talus under the entrance. In locations, infills become cemented as they receive drip from calcium carbonate rich water that has passed through the dolomite. And at times and in some locations, slightly acidic groundwater can decalcify such previously cemented infills, creating a soft matrix (Kuman and Clarke 2000). The infills were formed by surface material eroding into the caves and not by hominids depositing the material. The caves themselves were never used as living sites (Kuman and Clarke 2000). The Sterkfontein Caves captured a time period from about ~3.5 Ma to ~113,500 years ago (Partridge 2000, Reynolds, Clarke and Kuman 2007). The breccia deposits are divided into Members (M), ranging from the oldest (M1) to the youngest (M6). M5 is the only member which has yielded tools, although the Post-M6 Infill has mid to late Pleistocene artefacts. The other members only contain fossilised bone and natural stone, although wood is preserved in M4. Tools from M5 were analysed in this study and therefore I focus in following paragraphs on the environment and sediment conditions preceding and during this infill.

During the M4 infill the environment was mixed woodland with open components. It was drier and warmer than present day and the seasons were less pronounced. There is a lapse of time between M4 and M5, which is accompanied by erosion (Kuman and Clarke 2000, Partridge 2000, Hopley *et al.* 2007). Because M4 was deposited before the end of a period of global cooling and aridification, it is likely that this intermediate period was cooler and more arid than when M4 was deposited (Kuman and Clarke 2000, Partridge 2000, Hopley *et al.* 2007).

StW53 Infill (formerly Member 5 West (Kuman and Clarke 2000, Partridge 2000, Hopley *et al.* 2007) is the oldest non-tool bearing deposit in the M5 area. Because this study uses stone tools, I focus only on the artefact bearing deposits. Member 5 East-Oldowan (M5E-O) contains Oldowan artefacts that are older than the Member 5 East-Acheulean (M5E-A) deposit, which contains younger Acheulean tools (Kuman and Clarke 2000). I analysed tools from the Acheulean period, but because Loy analysed some tools from the Oldowan deposit I also included an overview of M5E-O. The pH in M5 varies between 8.34 and 10.38 (Jones 1998); the environment in the cave was alkaline.

Member 5E-O

This deposit is dated to ~2.0-1.7 Ma old (Kuman and Clarke 2000). The M5E-O deposit is dark reddish brown, poorly calcified clayey sand and contains abundant chert and dolomite and occasional faunal remains (Partridge 2000). The bone was accumulated mainly as a death trap assemblage, and in small part through slope wash around the cave entrance (Kuman 1994, Pickering 1999).

The average mean annual precipitation is thought to have been 477 millimetres (range 285-669 millimetres), of which 25% would have been winter rainfall. The average minimum monthly temperature was 8°C (range 4.7-10.2) and the average maximum monthly temperature was 31°C (range 29.1-33.7) (Avery 2001). Micro-mammals indicate an open habitat with medium density woodland or bush and grass along the river. There is a component of moist savannah (Avery 2001). Fossil macro-mammal remains shows some increase in the open habitat elements in Member 5E-O (Kuman and Clarke 2000), but Luyt and Lee-Thorp (2003) and Kuman and Field (press) interpret a moderately wooded habitat at this time. There is a general oscillation trend in this period, where arid adapted vegetation (C4 grasses) increase (Hopley *et al.* 2007, Lee-Thorp, Sponheimer and Luyt 2007).

Member 5E-A

This deposit is estimated by fauna to ~1.5 Ma old (Kuman and Clarke 2000). This member is yellowish red, well-calcified silty sand with less abundant chert and dolomite rubble (Partridge 2000). The fossilised faunal remains are fragmentary and are accumulated by hominids, predators and weathering processes (Partridge 2000); (Pickering 1999). The average mean annual precipitation was thought to have been 533 millimetres (range 285-781), of which 23% was winter rainfall. The average minimum monthly temperature was 5°C (range -0.1-4.3) and the average maximum monthly temperature was 31°C (range 29.1-33.7) (Avery 2001). Although precipitation was higher than during the M5E-O phase, the effective rainfall could have been less due to increased summer rains (Avery 2001). Oxygen and carbon isotope analysis indicate a rapid oscillation and opening with C4 grasses of the environment from 1.7 Ma ago onwards (Hopley *et al.* 2007, Lee-Thorp, Sponheimer and Luyt 2007, Luyt and Lee-Thorp 2003). Micro-mammals imply that the environment opened up even more compared to the previous deposit and that the

moist savannah component decreased, but by no means disappeared (Avery 2001). Faunal remains also indicate a shift towards a dry, more open grassland habitat (Kuman and Clarke 2000).

The environment during tool deposition

Brain (1995) suggests that the cave infills from the Cradle of Humankind sites, which include Sterkfontein and Swartkrans, were deposited during the onset of interglacial periods. The lack of vegetation and the increased rainfall permitted erosion. However, as Avery (2001) points out, rainfall towards the end of an interglacial period is also much higher than during a glacial, and by then vegetation is again reduced by deteriorating climate. Consequently erosion can take place. Pickering *et al.* (2007) demonstrate that the clastic deposits at Gladysvale, also one of the Cradle site, derive from cooler and more arid periods, compared to the present day climate, with stormy events leading to erosion (although this is not necessarily linked to glacial-interglacial cycles). In general the members were deposited in periods of less vegetation or declining vegetation.

M5E-O deposits are clayey sand and M5E-A is silty sand. If at the time of deposition the climate deteriorated and erosion became more pronounced one would generally expect the deposits to contain larger and less rounded sediment particles. Sand is larger and less rounded than silt and clay respectively. Deposition of the clastic sediments (breccia) was erratic and probably occurred during stormy and bad weather events (Pickering *et al.* 2007). Clay and silt are formed under pedogenesis outside the cave. The clay and silt from the sediments in M4E-O and M5E-A indicate that outside the cave, before erosion, palaeosols were present (also see (Kuman and Field in press)). Because M5E-A contains slightly less rounded particled than M5E-O, it is likely that the pre-M5E-A palaeosol was less stable and erosion may have been more of an ongoing process than during M5E-O. Manganese stains on tools from the nearby Coopers site possibly indicate that the tools were exposed for thousands of years. The Sterkfontein artefacts also have manganese stains and perhaps this indicates prolonged exposure. However, decalcification may also cause staining and more study is needed (Pickering pers. comm. 2008). It is probable that the stone tools from M5 lay at the surface around the caves for long periods and only washed into the caves when vegetation decreased and precipitation increased. In a tentative senario the

tools may have been deposited during the interglacial and washed into the cave as the interglacial ended.

During the M5 infills the climate was warmer and slightly drier than present day. The clay and silt in the sediment matrices indicate a stable palaeosoil outside the cave. The tools were deposited on these stable soils and then washed into the cave. Jones (Jones 1998) measured the pH of the breccia in M5. Breccia is clearly alkaline and ranges in pH from 8.34 to10.38.

The experimental settings

The experimental tools, discussed in Chapter Two, were deposited around a modern cave entrance and on the rubble slope under the shaft entrance, inside the cave. The sediment outside the cave has an average pH of 7.2. Inside the average pH is 7.8 (Table 3.9). The soil water content at Sterkfontein was measured with gypsum blocks (Eijkelkamp soil moisture reader model 14.22). The gypsum blocks were placed at four stations at the site (see Figure 2.2). After the blocks reached equilibrium I measured soil moisture tension in Pa (see Table 3.9). Although gypsum blocks are a common tool to measure soil moisture, this method only gives broad results. A measurement of >100-100 Pa implies that a soil is oversaturated; a reading of 95-100 Pa indicates the soil or sediment is at field capacity*; a measurement of 79-75 Pa is the required range for crop growth; a reading lower than 75 Pa implies that there is insufficient moisture for plant growth. The soil outside the Sterkfontein Cave is moist and at field capacity, but the soil inside the cave is much drier. The Sterkfontein soil samples are not hydrophobic like the Sibudu samples, indicating that although the soil from inside the cave is dry, it does occasionally come in contact with water.

The soil today at Sterkfontein is rich in organic plant materials, for example, rootlets, charcoal, pollen, insect remains and plant fibres (Figures A.97-A.100). The soil both in and outside the cave is brown sandy clay and a large portion consists of humus. The micro-remains are, like the quartz grains, reasonably large and therefore

* Field or storage capacity is a term used in soil sciences. It indicates the amount of soil moisture present after excess water has drained away and when downward water movement stopped. This is generally achieved in no more than two to three days after rain or irrigation. Fine textured soils hold more water and hold it more tightly (Berendsen 1997; Strahler & Strahler1997).

easy to identify (Figure A.99). The sediment also contains some bluish bone-like flakes (Figure A.101) and large amounts of hyphae (Figure A.102).

The Sterkfontein breccia is different from the modern sediment in that it is compact and it contains no organic micro-remains (Figure A.103). In the red breccia fossilised bone and small rubble clasts are present. Black manganese deposits visible on the broken breccia surface (Figure A.104). and fat-like deposits are present (Figures A.105-A.107).

Table 3.10: Soil moisture content and pH values at the various measuring station at Sterkfontein and the average pH from the M5 breccia.

Station	Moisture reading in Pa	pH value
1 (inside cave)	28	7.8
2 (inside cave)	16	-
3 (outside cave)	85	7.2
4 (outside cave)	79	-
Breccia M5 (average)	-	9.36

The present sediments around Sterkfontein cave entrances are moist and inside the caves the sediments are much drier. Outside the cave microbial activity is high, but inside it is low. In ancient times tools lay around the cave entrances for extended periods. Although the environment was generally drier, there must have been significant bioactivity in soil around the entrances. During the erratic deposition events of the clastic sediments, the caves were moist. However, it is likely that the caves were relatively dry when there was no deposition. For an overview of the past and present environmental settings at Sterkfontein see Table 3.14.

I expect residues to preserve inside the cave. Here the dry and sheltered environment, which has little bioactivity, protects residues. It is definitely suitable for plant tissue preservation and likely to be suitable for starch, resin, muscle, blood, bone and fat preservation. In addition, the slightly alkaline sediments, in combination with the dryness inside the cave, are favourable for residue preservation. Outside the cave, where soil moisture is fluctuating and pH is around neutral, no organic residues will preserve, but perhaps water-insoluble resin could. Microbial activity is high here and the elements affect the residues.

Although in ancient times the clayey component in M5E-O could preserve proteins (Jones 1998) and starch inside the dry cave, this is not the environment in which the tools were originally deposited. Jones (1998) demonstrates how blood residues could

preserve on ancient Sterkfontein tools. She suggests the following post-depositional process of ancient Sterkfontein residues (Jones 1998, 102-103):

1. Blood dries onto a stone tool surface
2. The stone tool is incorporated into sediment and comes in contact with clay
3. Sheet washing of sediments redeposits the soil as the infill breccia. Clays forming within the caves add to the overall clay content of the breccia.
4. Leaching of the dolomite bedrock by carbonic acid from rainfall releases calcite into solution.
5. Calcite ions form ionic bonds with clay particles.
6. Continued leaching of dolomite and the cycle of intermittent wetting (movement of water table) and drying precipitates micro and macro euhedral or anhedral crystals.
7. Calcification compacts the clay layers, which impinges on the residues.
8. Subsequent calcification(s) act to further protect blood residues by preventing water from coming into contact with the proteins.

However, some of the points she makes are not relevant to deposition of tools at Sterkfontein and, as a result, they are perhaps even incorrect. First, it is unknown if blood on tools was able to dry before deposition in the soil. Dry blood has a better survival chance (paragraph 3.2.4.2), but even if the residues dried before deposition, it is likely that they were rehydrated in the moist soil. Secondly, only sediments with high clay content shield residues from microbial activity (see paragraph 3.2.2). Although the clay content is high in the M5E-O deposit, the residues were not protected while the tools lay outside the cave on the humic layer of the palaeosol. Points 4-8 argue that the residues were protected by a compacted clay and calcite microenvironment. However, by the time these processes became relevant, the tools had already been exposed to the elements for an unknown period. The tools were deposited outside the cave where microbial activity was high and where the elements had free rein on the tools and residues. The tools may have lain outside the cave for a considerable period of time, where the protective clay-calcite process does not take place. Note that the tools in M5E-A were deposited in silty sand sediment, which is more destructive to organic materials than clay sediments. I predict that no ancient organic residues are preserved on the Sterkfontein tools.

3.3.2 Sibudu Cave

Sibudu Cave is located about 40 kilometres north of Durban, KwaZulu-Natal province, South Africa. The cave, which is more a shelter, is situated 15 kilometres inland from the Indian Ocean on a steep cliff overlooking the Tongati River (Wadley and Jacobs 2006). The shelter has been worn out by fluvial action from the prehistoric Tongati River in a Natal Group sandstone cliff. Sibudu is located about 100 metres above mean sea level. Current summer rainfall averages 750 millimetres and winter rainfall 250 millimetres. Sibudu summers are humid and hot with mean January temperatures between 22-25°C. Winters are cool and drier with mean July temperatures between 17-20 °C. Although the forest is mostly cut now, Sibudu falls in a woodland zone with mainly evergreens, but also some deciduous and semi-deciduous trees.

The site is in near primary context (Pickering 2006) allowing climatic, environmental and activity reconstructions as well as extensive stone tool analysis between time periods (*e.g.* Allott 2006, Cain 2006, Delagnes *et al.* 2006, Lombard 2006a, Reynolds 2006). At Sibudu, bone remains and stone tools are found in abundance and the stratigraphic layers in which they are found are dated from the final Middle Stone Age (MSA) to a pre-Still Bay age (~37 ka to older than 75ka) (Wadley and Jacobs 2006) and fall within OIS 4 and 3. Thus far no macro plant remains have been found, other than charred seeds. The tools in this study come from Howiesons Poort layers and are dated to >60 ka (Wadley and Jacobs 2006).

OIS 4 (75-64 ka) was a cool period during which the sea levels dropped by -60 to -70 metres. OIS 3 (64-32 ka) was a relatively warm interstadial. During OIS 3 sea levels fluctuated between -40 and -70 metres. Both periods were generally cooler and drier than present day and sea levels never reached present levels (Wadley 2006).

Clarke *et al.* (2003, but also see Botha *et al.* 1992, Botha 1996, Botha and Partridge 2000, Clarke *et al.* 2003, Munyikwa 2005) reconstruct prehistoric environmental and climatic settings for the Late Quaternary of northern KwaZulu-Natal by investigating and dating periods of colluviation and pedogenesis. During colluviation, extensive erosion takes place; hillslopes become covered in sandy hillwash sediments and gully erosion intersects these deposits. Colluviation is associated with aridity, reduced vegetation and transitional climates. Pedogenesis indicates more humid conditions with more vegetation during which palaeosols

formed (Clarke *et al.* 2003). During OIS 4 the episodes of aridity were more extreme than during OIS 3 and by OIS 3 the fluctuations diminished, but it remained generally drier and cooler than present day (Cohen *et al.* 2007, Scott, Holmgren and Partridge 2008). Clarke *et al.* (2003) identify several periods of colluviation, namely from 110-96 ka, from 56-52 ka, from 47-41 ka and around 11 ka. These episodes concur with data from pollen and data from the Lake Malawi drill cores (Cohen *et al.* 2007, Scott, Holmgren and Partridge 2008). The dry periods correspond with hiatuses in the Sibudu stratigraphy (Wadley 2006), indicating that the Sibudu material was deposited in periods of pedogenesis and therefore during warm and humid times, relative to the periods of colluviation. This also means that during drier periods no sterile, geological layers formed at Sibudu and the subsequent sedimentation (mostly anthropogenic in nature) only occurred during warmer/humid times.

Many of the plant taxa found presently around the cave, we also find in charcoal from analysis and seeds. Some of the prevailing conditions were probably similar. Some taxa, however, do not currently grow in coastal regions and it is likely that dry woodland vegetation was more abundant then.

All through the sequence, riverine woodland vegetation and bushveld or more open vegetation was present. However, the taxa changed over time indicating large-scale climatic changes (Allott 2006). Climate and environment can be reconstructed with charcoal, seed, bone and magnetic analysis and in the paragraph below the following one I discuss the sediment analysis (Allott 2006, Cain 2006, Glenny 2006, Herries 2006, Reynolds 2006, Sievers 2006):

- Pre-60 ka Howiesons Poort layers: Presence of evergreen woodland. Rainfall was relatively high or there was high moisture availability in the soils, from the river or from humidity levels. Temperatures were cooler than present day.
- Howiesons Poort layers: Habitat was drier than present. Perhaps it was also warmer than present day or receiving more sunlight. Vegetation was similar to present vegetation in northern KwaZulu-Natal in woodland and bushveld communities.
- ~60 ka layers: More riverine and woodland taxa were present than in previous periods. Less dry bushveld vegetation occured, indicating a riverine woodland/evergreen woody community. Some taxa indicate cold winters with frost. Faunal remains indicate drier conditions than present with open

woodland vegetation. The early parts were cooler and there was a general warming trend.

- ~50 ka layers: Deciduous taxa were more prominent in this period than in other periods. More bushveld taxa indicate dry and warm bushveld and savannah communities. However, woodland and riverine indicators were still present in relatively the same numbers as the previous period. The landscape probably became more open. The faunal remains indicate that the environment was drier than present with open woodland vegetation. There appears to have been a general warming trend.
- ~37 ka layers: Some of the taxa from these layers are currently found near the shelter. Conditions were probably similar to the present, but drier.

In general, in the periods during OIS 3 when Sibudu was occupied, the climate was drier and somewhat cooler than present.

The up to three metre deposits at Sibudu Cave are anthropogenic and display a micro-stratigraphy at a centimetre scale (Pickering 2006). Deposits consist of silty ash with no visible sand fraction; the angular fragments of bone and charcoal show no sign of water working and are anthropogenic. The layers show no clear erosion boundaries and they are consolidated and somewhat hard (Pickering 2006). At Sibudu many clearly visible hearths are present and a significant number of stone artefacts show signs of exposure to heat (Wadley 2006).

The absence of phosphate minerals in the Sibudu deposits indicate that diagenesis of sediments is slow (Schiegl and Conard 2006). Gypsum is a secondary hydrated calcium mineral, which is present in the Sibudu deposits. Gypsum needs moisture to grow, but is also soluble when more water is available (Schiegl and Conard 2006). In the ~50 ka and ~60 ka layers more gypsum is present than in the other layers above and below. This indicates that the formed surfaces were stable enough to allow gypsum to develop, but were also covered rapidly to prevent further gypsum growth beyond ~2 centimetres (Schiegl and Conard 2006). This concurs with environmental data above. The pre-60 ka periods were more humid. There is little to no gypsum in the deepest layers (where the tools from this study are from). It is possible that the fewer hearths in these levels did not supply enough sulphite for gypsum formation. Another explanation is that somewhat higher moisture levels at these deep deposits dissolved gypsum (Schiegl and Conard 2006). Considering the

relative high humidity of the pre-Howiesons Poort and Howiesons Poort layers, it appears that gypsum dissolved again in these levels.

In Sibudu's sedimentary milieu, bone dissolution is minimal and bone preservation can be expected. However, the crystal gypsum growth mechanically weathers bone (Schiegl and Conard 2006) and bone was less well preserved in the 60 and 50 ka sediments than the pre-60 ka sediments. A significant part of the phytoliths at Sibudu suffered from chemical attacks while they were exposed to high alkaline conditions (pH 9-13.5) in prevailing plant ashes (Schiegl and Conard 2006). This was probably caused during burning. Ash quickly becomes more acidic after deposition and Williamson (2004) and I measured much lower pH values for the ancient sediments (see Table 3.11). The ~ 50 ka levels have an average pH of 7.40; the ~ 60 ka levels have an average pH of 6.75; the >60 ka levels have an average pH of 7.08. I observed that the ancient sediments, which I measured are hydrophobic; meaning that when I added water to a sediment sample, lumps of sediment floated on the water and it took considerable shaking to mix the water and sediment. This indicates that the sediments are dry and have been for a considerable time.

Table 3.11: Overview of the pH values of selected sediment units from Sibudu. GL= Geeske Langejans; BW= Bonny Williamson.

Sediment unit	Period	pH	Analyst
Orange Mottled Deposit	~ 50 ka	7.64	BW
Oreo	~ 50 ka	7.37	BW
Red Speckled	~ 50 ka	6.41	BW
Brown Speckled	~ 60 ka	7.10	BW
Chocolate-Spotty Camel	~ 60 ka	6.40	BW
Grey Rocky-Spotty Camel	~ 60 ka	5.90	BW
Speckled sunrise	~ 60 ka	5.53	BW
Mexican Yellow	~ 60 ka	5.56	BW
Mustard	~ 60 ka	5.79	BW
Chocolate 2	~ 60 ka	5.49	BW
Brown Grey Mix 2	~ 60 ka	5.45	BW
Grey Rocky	>60 ka	6.54	GL
Grey Rocky II	>60 ka	6.88	GL
Hearth in Grey Sand II	>60 ka	7.27	GL
Grey Sand II	>60 ka	7.30	GL
Pinkish Grey Sand	>60 ka	7.43	GL
Average pH all levels		6.50	
Average pH ~ 50 ka levels		7.14	
Average pH ~ 60 ka levels		6.75	
Average pH >60 ka levels		7.08	

The >60 ka layers were deposited during humid and warm, yet drier and cooler than present-day circumstances. Sedimentation rates were rapid, meaning that the tools were covered quickly. Almost no diagenesis took place and bones are well preserved.

The stone tools are made from local dolerite and occasionally sandstone. Dolerite river cobbles and rare hornfels, quartzite and quartz river nodules are also used for knapping. Hornfels, quartzite and quartz probably have a richer source further away from the shelter (Wadley and Jacobs 2006).

Williamson (2004, 2005) and Lombard (2004, 2005, 2006a, 2006b, 2008) conducted residue analysis on stone tools from Sibudu Cave. Both found plant, animal and inorganic (*e.g.* ochre) remains on artefacts from different stratigraphic layers. Lombard uses residue analysis to demonstrate hafting of Howiesons Poort segments and post-Howiesons Poort points (2006a, 2006b).

The experimental tools, discussed in Chapter Two, were deposited near the dripline and far into the interior of the shelter. The sediment around the dripline has an average pH of 6.5. Inside the average pH is 7.2 (Table 3.10). These values are similar to the values from the ancient sediments. The soil water content at Sibudu was measured with gypsum blocks (Eijkelkamp soil moisture reader model 14.22). The gypsum blocks were placed at four stations at the site (see Figure 2.3). After the blocks reached equilibrium I measured soil moisture tension in Pa (see Table 3.12). Although gypsum blocks are a common tool to measure soil moisture, they only give broad results. A measurement of >100-100 Pa implies that a soil is oversaturated; a reading of 95-100 Pa indicates the soil is at field capacity; a measurement of 79-75 Pa is the required range for crop growth; a reading lower than 75 Pa implies that there is insufficient moisture for plant growth. The sediment around the Sibudu dripline is dry, but the sediment inside the shelter is extremely dry. I also observed that the sediment from inside the shelter is hydrophobic. The topsoil used in the experimental setting and the archaeological sediment samples had a hydrophobic response. This indicates that the Sibudu sediment is dry and has been for considerable time.

Table 3.12: Soil moisture content and pH values at the various measuring stations at Sibudu.

Station	Moisture reading in Pa	pH value
1 (dripline)	16-17	6.4
2 (dripline)	8-9	6.5
3 (interior)	5	7.3
4 (interior)	5	7.1

The present and old sediments at Sibudu are dry; there is no precipitation that can lead to diagenesis. Microbial activity inside the shelter is low. No earthworms or other organisms are able to live in this dry environment, which is also not rich in easily accessible nutrients. In ancient times, tools were rapidly covered under newly deposited sediments. Consequently, they were only exposed to the humid environment for a short time. Towards the dripline, the sediment becomes more humid and organic and microbial activity is higher.

Inside the cave the ashy sediment contains microscopic burnt plant remains (Figures A.108 and A.109). Burnt plant remains vary in colour from white to black (Figure A.110). Microscopic burnt fatty bone particles are also present. These fatty flakes are yellow and orange in colour. Larger bone pieces are also abundant and these pieces can be fairly fatty; they are yellow to orange in colour. Few pollen, spores, hyphae and insect remains are present (Figures A.111 and A.112). The sediment at Sibudu is similar inside and outside the shelter. The humus content of the soil matrix becomes higher towards the dripline; larger unburnt plant remains are more abundant (Figure A.113). The ashy matrix at Sibudu is fine-grained and obscures starch identification. Under the microscope ash is yellow and orange, but sometimes also white and grey. The last two colours are problematic as they are similar to fat. Ash has the opaque properties of fat in cross-polarized light. When smeared over the surface, it can mimic fat deposits (Figure A.76). The soil in the >60 ka layers is similar to the topsoil (Figures A.114 and A.115). Plant remains are slightly scarcer and burnt charcoal remains are more abundant. These layers also contain fat-like remains (Figure A.114) and burnt fatty bone remains (Figures A.116 and A.117).

Because Sibudu is in primary context, the environmental reconstruction can be used to make predictions regarding residue preservation at this site. Because of the extreme dryness of the Sibudu site, residues are expected to preserve inside the shelter. Because the dripline area is more humid and has more microbial activity, residues are not expected to preserve or least not as well as inside the cave. Because there are

many hearths, burnt tools and sediment particles, I expect to find burnt use-related residues in the archaeological sample. For an overview of the environmental settings at Sibudu, past and present see Table 3.14.

I expect no muscle tissue, blood, starch or plant tissue preserved in the Sibudu experiments outside the shelter. Perhaps resin, bone and fat might preserve, but it is unlikely that they could survive. I expect the residues from the Sibudu experiments inside the shelter to preserve better. Resin might preserve, but the cave could also be too dry and the resin might have crumbled. It is likely that muscle tissue, blood, fat and bone should preserve. In the Sibudu >60 ka archaeological sample it is likely that plant tissue and starch preserve. It is possible that resin and fat survive and likely that muscle tissue, blood and bone preserve.

Because drier or less humid conditions prevailed in later times, I expect that residues preserve better in the ~60 ka, ~50 ka and ~37 ka layers than in the >60 ka layers.

3.3.3 Wilhelmina Polder

The Wilhelmina Polder is located in the South West of The Netherlands, in Zeeland province. The area is part of an agricultural cooperation that uses the land mainly to grow cereals. The soil consists of heavy marine clay with few macro plant remains[*]. The carbon cycle at Wilhelmina Polder is rapid, meaning that microbes quickly decompose organic remains. The soil is not extremely permeable and when moist the clay swells, holding the water. This reclaimed land has no natural vegetation. Besides large agricultural areas, there is some cultivated grassland with a few deciduous trees.

Mean annual precipitation is 721 millimetres, which falls all year round; the monthly minimum temperature is 3.7°C and the maximum monthly temperature is 17.8°C (KNMI 2002). For an overview of the environmental settings at Wilhemina Polder see Table 3.14.

I did not measure the soil moisture contents at Wilhelmina Polder. However, because intensive agriculture is possible and due to the high rainfall and low evaporation rates in this area, soil moisture must be high and at least at field capacity. The pH at Wilhelmina Polder is 7.40 (Table 3.13) and is therefore slightly alkaline.

[*] Information available on http://www.bodemdata.nl/

Table 3.13: The pH values of the sediments at Wilhelmina Polder and Zelhem.

Measure station	pH
Wilhelmina Polder	7.40
Zelhem humus top layer	3.94
Zelhem sediment	3.87

The clay is fatty in appearance with small particles that easily disguise use-related residues (Figures A.118 and A.119). Although clay ranges in colour from grey to black and blue, under the microscope the colours appear much lighter. Fat can also appear light-bluish. Many residues take on the fatty properties from clay, possibly leading to misidentification (Figure A.120). The clay sediment contains few organic remains. There are some plant remains, pollen, hyphae and spores in the sediment matrix (A.121 and A.122). Most plant material is white in colour.

I expect poor residue preservation on the experimental sample from Wilhelmina Polder. It is unlikely that plant tissue or fat can preserve; but it is possible for starch and bone, but not impossible for resin, muscle tissue and blood.

The bioactivity in the soil is high as is the precipitation and both have a negative effect on residues. There are no special circumstances that could lead to residue preservation, such as presence of heavy metals in the sediment. The sediment is slightly alkaline, but due to the moisture this has no preserving effect. Although clay particles can lock and thus preserve micro-remains and molecules, the sediment at Wilhelmina Polder is not compacted and, due to frequent ploughing, it is quite loose. This loose sediment probably preserves remains less well than compacted clay sediment. However, due to the preservation qualities of clay, I expect more residues to preserve in this clay sediment than in the sandy sediment at Zelhem.

3.3.4 Zelhem

Zelhem is located in the East of The Netherlands, in Gelderland province. The area is used for agriculture. The soil consists of sand and the macro plant component is moderate for the topsoil in which the tools were buried[*]. The carbon-cycle at Zelhem is rapid, meaning that microbes quickly decompose organic remains. The sandy

[*] Information available on http://www.bodemdata.nl/

sediment is permeable and, when moist, water quickly drains away. The natural vegetation is deciduous woodland with open components.

Mean annual precipitation is 758 millimetres, which falls all year round; monthly minimum temperature is 2.1 °C and the maximum monthly temperature is 17.0°C (KNMI 2002). I did not measure the soil moisture contents at Zelhem. However, because agriculture is possible and due to the high rainfall and low evaporation rates in this area, soil moisture must be high and at least at field capacity. The humus top layer is 3.94 and the sediment under the top layer is 3.87 (Table 3.13). This makes the soil at Zelhem acidic. For an overview of the environmental settings at Zelhem see Table 3.14.

The Zelhem sediment consists of large quartz particles (Figure A.123). The quartz grains can appear similar to hyphae-damaged starch. The sand matrix contains abundant sediment organics (Figures A.123 and A.124). Larger and smaller plant particles occur, ranging in colour from yellow to dark brown. Hyphae, spores, insect remains and pollen are also plentiful (Figures A.125 and A.126). Except for damaged starch, the sediment and sediment organics are easily distinguished from use-related residues.

I expect no organic residues to preserve at Zelhem. The bioactivity is high and so is the precipitation and both these attributes are bad for residue preservation. The sediment is permeable and acidic and this increases chemical weathering. There are no special circumstances that could lead to residue preservation, such as presence of heavy metals in the sediment. I expect that fewer residues preserve on the Zelhem artefacts than on the Wilhelmina experiments. The clay minerals could protect the organic remains, but because at both sites the bioactivity and precipitation are high and because the soil structure is loose, the difference will be minimal.

3.3.5 Site synthesis

The four localities and the different time phases described above are significantly different from each other, which is why they were selected for this project. Table 3.14 present an overview of the environmental and sediment conditions at each site and time phase. Good preservation of organic residues is expected inside the modern day Sterkfontein Caves and Sibudu Cave and in the ancient Sibudu >60 ka sample. The

remaining experimental and ancient settings are not expected to preserve residues. From the variables, which influence decay, moisture appears to be the most important one in these experiments. The pH is relatively neutral at all sites, with the exception of Zelhem, and it appears that moisture is the variable that is different at the sites and could account for predicted varying preservation. The acidity of the Zelhem sediment only increases the decay. The combination of Figures 3.2 to 3.9 with Table 3.14 leads to the following predictions for the samples inside modern day Sterkfontein Caves and Sibudu Cave and in the ancient Sibudu >60 ka sample: plant tissue, starch, muscle tissue, blood and hair are expected to have a good to intermediate preservation in dry and neutral to slightly alkaline environments. Resin and fat will not preserve as well in these circumstances, but bone is expected to have a good preservation.

Table 3.14: Overview of environmental and sediment conditions at the studied sites. X = existing or reconstructed circumstances; - = not applicable; N.i. = no information available. When a site has two crosses divided over the alkaline, pH neutral and acidic columns, it means that the sediment is slightly alkaline or slightly acidic.

Site	Wet Soil	Dry Soil	Atmosphere dry	Atmosphere humid	Alkaline	pH neutral	Acidic	Heavy metals	Sediment type	Soil constitution	Oxygen depleted	No UV light	Temperature
Sterkfontein outside cave	X	-	-	X	-	X	-	N.i.	Sand/clay	N.i.	-	-	Moderate
Sterkfontein inside cave	-	X	-	X	X	X	-	N.i.	Sand/clay	N.i.	-	Less light	Moderate
Sterkfontein M5 before washing in	X	-	-	X	N.i.	N.i.	N.i.	N.i.	Clay/silt	N.i.	-	-	Moderate
Sibudu outside shelter	X	-	-	X	-	X	X	N.i.	Silt/ash	N.i.	-	-	Moderate
Sibudu inside shelter	-	X	-	X	-	X	-	N.i.	Silt/ash	N.i.	-	-	Moderate
Sibudu >60 ka	-	X	-	But drier than today	-	X	-	N.i.	Silt/ash	N.i.	-	-	Moderate
Wilhelmina Polder	X	-	-	X	X	X	-	N.i.	Clay	Less permeable	-	-	Moderate
Zelhem	X	-	-	X	-	-	X	N.i.	Sand	Permeable	-	-	Moderate

4 RESULTS FROM THE REPLICATED SAMPLE

This chapter presents the results from the experiments. Stone flakes were used to process a variety of materials to generate starch, woody plant tissue, muscle tissue, bone, fat and blood residues. I deposited these flakes at Sterkfontein and Sibudu, South Africa, and in clayey and sandy sediment at Wilhelmina Polder and Zelhem in the Netherlands respectively. The residue bearing tools were exposed to a variety of conditions to understand how these conditions influence residue preservation. I am interested in the influence of time, residue type, sediment and climate/environment on residue preservation. In addition, sandpaper slides of different grain sizes were set out with blood, fat and starch residues to investigate the influence of the stone material and micro-environments on residue preservation. These slides were deposited on clayey sediment Willemina Polder and at Zelhem in the Netherlands. All experiments have a control sample of unused flakes, the blanks. From the blanks I want to learn if sediment organics adhere to artefacts and in what quantity. In addition, I want know if sediment organics indeed leave a random distribution pattern on the tool's surface or not. In addition to the sediment samples, discussed in Chapter Three, the blanks give insight into the types of sediment organics present at the sites and their visual characteristics.

I discuss the results using tables, micrographs and distribution maps. The micrographs and maps are presented in Appendix B. I start by discussing the data from the blanks. The second half of this chapter discusses the results of the replicated tools. The results are presented in tables with a preservation index (PI) indicating how well residues are preserved; the index is introduced in Chapter Two. I summarise the different sets of experiments by site; in the Discussion and Conclusion in paragraphs 4.6 and 4.7, I deduce various universalities about residue preservation and analysis.

Residue analysis is not only constrained by the quantities of visible residues left, but also by factors like contaminants and raw materials. In Chapter Two I discussed the sites, raw materials and contaminants in detail; in the following paragraphs I implement these data on the blank control sample and the replicated sets.

4.1 THE BLANK CONTROL SAMPLE

All replicated tools, blanks and slides contain contaminants. Contaminants can come from handling or storage, but in this part of the project they came from the sediment. It is important to know what the possible contaminants are in order to exclude them from the analysis. Therefore I examined blanks and sediment samples. The main contaminants that are present on the blanks, are discussed in Chapter Two; in this section I review in what quantity sediment organics occur on blanks and if the remains present a random or meaningful distribution pattern. Tables 4.1 to 4.4 present an overview of the blank sample.

All micro-remains, which are part of the sediment matrix, can adhere to the tool surface. The contaminants on the artefact depend on what is present in the sediment matrix. There is no preference for particular types of remains. Plant residues and bone require special mention. These types are common elements in sediments. Plant tissue, plant fibres, starch grains, fatty bone, bone and connective fibres can be confused with ancient use-related residues. However, the distribution of sediment organics on the blanks is random. This is in contrast to the meaningful pattern that use-related residues leave on used tools.

Figures B.2 to B.17 present a selection of distribution maps from blanks from the Sibudu, Sterkfontein and Zelhem samples. The maps show no patterning of organic remains that indicates tool use. The remains are spread arbitrarily over the surfaces and have no preference for a particular area or edge. For example, Figure B.10 clearly illustrates the random distribution of plant fibres over the surface. Studying the distribution of remains can eliminate contaminants. Figures B.12 and B.13 demonstrate that all remains, including pollen, spores, hyphae, plant fibres and plant tissue exhibit this random spread.

In general, few residues adhere to blanks. The processed experimental tools, discussed in the next section, contain more sediment remains. Moist, use-related residues act as adhesive and therefore used tools hold more sediment remains than unused blanks.

Haslam (2006a) suggests that hyphae grow on use-related residues and they can help to locate the working edge and to reconstruct tool use. However, hyphae are

present in the sediment (Figures A.111 and A.126) and grow on all organic remains, including sediment remains (Figure B.18 and B.19). Hyphae are also present on blanks and therefore cannot be used to reconstruct tool use (Figure B.20).

On the Sibudu and Sterkfontein blanks, several confusing sediment remains are present. On the Sterkfontein blanks I found residues, which are similar to fat droplets (Figure B.21). Visually they are identical to fat droplets from the replicated sample and perhaps they are indeed fat. I also found a deposit that is similar to muscle tissue (Figures B.22 and B.23). Although it is unlikely that this is muscle tissue, I cannot identify it as yet. On the Sterkfontein blanks I also found fat-like remains. They have a bluish and opaque appearance (Figure B.24). On a Sibudu blank I found a fibre, which appears similar to a connective fibre (Figure B.25). Perhaps this is a plant fibre, which is damaged by hyphae. A few fatty bone-like deposits are also present (Figure B.26). On many blanks the burnt fatty bone deposits are present (Figure A.60). They are also abundant in the sediment matrix (Figures A.114-A.117). For an overview of general sediment particles, which are also present on the blanks, see Figures A.97-A.126. Although I am unable to identify all remains on the blanks, it is possible to distinguish them from possible use-related residues on processed tools. Some contaminants are obviously not use related. For example, the fatty green deposit in Figure B.27 contains pollen, which is not use-related.

Micro-remains can appear different on the stone surface than in the sediment sample (Figure B.28). The colour and texture of the stone influence viewing, and organic remains on tools are not always found in thick patches like sediment samples. Sediment organics can appear isolated on the tool and then they can also look different. I recommend setting out blanks at a selected archaeological site before commencing residue analysis. The sediment samples and the blanks give the analyst an idea of the type of contaminants present, how they look and furthermore it familiarises him/her with the rock types.

Table 4.1: Overview of the Sterkfontein blank sample.

Tool number	Raw material	Situation	Buried	Time
STK145	Chert	In cave	Exposed	1 year
STK146	Chert	In cave	Exposed	1 year
STK147	Chert	In cave	Exposed	4 weeks
STK148	Chert	In cave	Exposed	4 weeks
STK149	Chert	In cave	Buried	1 year
STK150	NOT RECOVERED			
STK151	Chert	In cave	Buried	4 weeks
STK152	Chert	In cave	Buried	4 weeks
STK153	NOT RECOVERED			
STK154	Chert	Outside cave	Exposed	1 year
STK155	Chert	Outside cave	Exposed	4 weeks
STK156	Chert	Outside cave	Exposed	4 weeks
STK157	Chert	Outside cave	Buried	1 year
STK158	NOT RECOVERED			
STK159	Chert	Outside cave	Buried	4 weeks
STK160	Chert	Outside cave	Buried	4 weeks

Table 4.2: Overview of the Sibudu blank sample.

Tool number	Raw material	Situation	Buried	Time
SB147	Hornfels	In shelter	Exposed	4 weeks
SB148	Hornfels	In shelter	Exposed	4 weeks
SB149	Hornfels	In shelter	Buried	1 year
SB150	Hornfels	In shelter	Buried	1 year
SB151	Hornfels	In shelter	Buried	4 weeks
SB152	Hornfels	In shelter	Buried	4 weeks
SB153	Hornfels	Dripline	Exposed	1 year
SB154	Hornfels	Dripline	Exposed	1 year
SB155	Hornfels	Dripline	Exposed	4 weeks
SB156	Hornfels	Dripline	Exposed	4 weeks
SB157	Hornfels	Dripline	Buried	1 year
SB158	Hornfels	Dripline	Buried	1 year
SB159	Hornfels	Dripline	Buried	4 weeks
SB160	Hornfels	Dripline	Buried	4 weeks

Table 4.3: Overview of the Willemina Polder blank sample.

Tool number	Raw material	Buried	Time
NLC 21	Flint	Exposed	6 months
NLC 22	Flint	Exposed	6 months
NLC 23	Flint	Buried	6 months
NLC 24	Flint	Buried	6 months
NLC 25	Flint	Buried	6 months

Table 4.4:Overview of the Zelhem blank sample.

Tool number	Raw material	Buried	Time
NLS 21	Flint	Exposed	6 months
NLS 22	Flint	Exposed	6 months
NLS 23	Flint	Buried	6 months
NLS 24	Flint	Buried	6 months
NLS 25	Flint	Buried	6 months

4.2 STERKFONTEIN

The variables tested at Sterkfontein are: time and its influence on residue preservation in a buried and exposed setting; the influence of a sheltered and unprotected environment; the influence of residue types on preservation. Ten sets of experimental tools are analysed. Every set has a blank control sample to help understand the chert and the post-depositional contaminants. I discuss the results per residue type and at the end of every section I summarise the results.

Unfortunately I lost the sample that was buried outside the cave. I replaced the chert tools with an identical set of similarly fine-grained quartzite experiments. Although the tool numbers below do not appear sequential, the experiments are perfectly comparable.

4.2.1 Starch sample

The results of the replicated and weathered starch sample are discussed in the following paragraphs. Tables 4.5-4.8 present the details of the experimental settings and the quantified preservation results.

STK81 and STK82: PI 2 and PI 2

Residues are present on both tools, mainly as single grains, but also in small patches. The residues are located around the working edges. A few small pieces of epidermal tissue are present. Hyphae are present, but they do not always occur in association with starch grains.

STK83 and STK 84: PI 4 and PI 4 (Figures B.61 and B.62)
Along the working edges thick and invasive starch deposits occur. Starch grains are always found in combination with hyphae. Black spores are also present. Red woody tissue is present along working edges. It is unclear if this tissue is a use-related or post-depositional residue.

STK85 and STK86: PI 2 and PI 2 (Figures B.63 and B.64)
A few isolated starch patches of starch grains occur around the working edges. These deposits are occasional and discontinuous. Some spore patches are present.

STK87 and STK 88: PI 3 and PI 3
Many starch grain patches are present and they are concentrated on the working edges of the tools. Thicker deposits of starch grains are located near the edge, but single grains are present all over the surfaces. Starch is always found in combination with hyphae.

STK89 and STK90: PI 0 and PI 0
Starch residues are absent. Some hyphae and spores are present along the edges.

STK91 and STK92: PI 2 and PI 2
A few starch grains are left on these samples. In both cases, one side is devoid of residues, while on the other side a few starch residues are present. There are only small accumulations and single grains present in cracks around the edge.

STK13 and STK14: PI 2-1 and PI 0 (Figures B.65 and B.66)
STK13: A few opaque starch grain deposits occur on this tool. The deposits are mainly concentrated on the working edges. Some epidermal tissue is present. Hyphae and sediment remains also occur.
STK14: Starch grains are absent on this tool. However many deposits of epidermal tissue occur. Some deposits are close to the working edge and others are more towards the centre of the tool.

STK15 and STK16: PI 1 and PI 3 (Figures B.67 and B.68)

STK15: Only one starch deposit on the dorsal side is preserved. The deposit contains deteriorated and opaque starch grains and hyphae. Pollen and hyphae are thin and equally spread over the surface and are not indicated in the distribution maps.

STK16: More starch grains are preserved on this tool than on STK15. They appear along the thin working edge and in several smaller starch deposits, away from the edge. Hyphae are growing on the grains and the starch grains are often opaque. Epidermal tissue is absent.

Table 4.5: Preservation index for the Sterkfontein starch experiments, deposited one year inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK81	Starch	In cave	No	One year	2
STK82	Starch	In cave	No	One year	2
STK85	Starch	In cave	Yes	One year	2
STK86	Starch	In cave	Yes	One year	2

Table 4.6: Preservation index for the Sterkfontein starch experiments, deposited one year outside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK89	Starch	Outside cave	No	One year	0
STK90	Starch	Outside cave	No	One year	0
STK13	Starch	Outside cave	Yes	One year	2-1
STK14	Starch	Outside cave	Yes	One year	0

Table 4.7: Preservation index for the Sterkfontein starch experiments, deposited four weeks inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK83	Starch	In cave	No	Four weeks	4
STK84	Starch	In cave	No	Four weeks	4
STK87	Starch	In cave	Yes	Four weeks	3
STK88	Starch	In cave	Yes	Four weeks	3

Table 4.8: Preservation index for the Sterkfontein starch experiments, deposited four weeks outside the cave

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK91	Starch	Outside cave	No	Four weeks	2
STK92	Starch	Outside cave	No	Four weeks	2
STK15	Starch	Outside cave	Yes	Four weeks	1
STK16	Starch	Outside cave	Yes	Four weeks	3

Starch is not well preserved at Sterkfontein. After one year most residues are entirely decomposed and when the cave does not protect the residues, four weeks are enough

to do considerable damage. Only the four-week experiments in the cave show reasonable preservation.

4.2.2 Wood sample

The results of the replicated and weathered wood tissue sample are discussed in the following paragraphs. Tables 4.9-4.12 present the details of the experimental settings and the quantified preservation results.

STK97 and STK98: PI 4 and PI 4 (Figures B.96 and B.70)
Many large and sometimes invasive clusters of plant tissue and plant fibres occur on these tools. Most are confined to the working edge. Some residues are visible with the naked eye. A few spots of possible resin are present. Thin hyphae strands are present all over the residues, but there are no spores.

STK99 and STK100: PI 5 and PI 5 (Figures B.71 and B.72)
Many large and sometimes invasive clusters of plant tissue and plant fibres are present. Most, however, are confined to thin working edges. Small starch grains are visible around exudates (on STK99) and plant tissue deposits. A few hyphae are present, but spores are absent. The fibres away from the edge are probably contaminants as are the rootlets on this tool.

STK101 and STK102: PI 3 and PI 3
Thin deposits of macerated plant tissue are found along the working edges. The deposits are not invasive. The plant deposits on these buried samples are darker in appearance than the plant deposits on the unburied counterparts. Because I found no hyphae or spores I do not think the deposits were degraded, but rather that they were smaller to begin with.

STK103 and STK104: PI 5 and PI 5
Along the edges of these tools an almost continuous patch of plant fibres and plant tissue is present. The residues are not invasive and are restricted to the edges. Some hyphae are present, but not always in association with the wood residues.

STK105 and STK106: PI 2 and PI 2

A few patches with plant tissue are preserved. The plant tissue is mainly present along the edges. Many hyphae are present along the edges and they occur in association with plant tissue. On both tools one of the two sides preserves more residues than on the other side.

STK107 and STK108: PI 2 and PI 2

A few sparse patches of plant tissue occur. Few hyphae and some small clusters of spores are present. On both tools, one of the two sides preserves more residues than the other sides.

STK29 and STK30: PI 2 and PI 3 (Figures B.73 and B.74)

Some plant tissue deposits are preserved on STK29 and STK30. The deposits are concentrated along the working edges. Rootlets and hyphae are abundant on STK30. The dorsal side of STK30 shows no residues, but on the ventral side several deposits are present.

STK31 and STK 32: PI 3 and PI 3-4 (Figures B.75 and B.76)

Several plant tissue deposits are present on these tools. The remains are concentrated near the working edges, but on STK32 the residues also occur in the interior of the surface. STK32 shows green colouration along the edges and this is probably plant exudate. Under high magnifications the stained surface appears indistinguishable from the normal surface. Some hyphae are present, but they do not cover the wood residues.

Table 4.9: Preservation index for the Sterkfontein wood experiments, deposited one year inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK97	Woody tissue	In cave	No	One year	4
STK98	Woody tissue	In cave	No	One year	4
STK101	Woody tissue	In cave	Yes	One year	3
STK102	Woody tissue	In cave	Yes	One year	3

Table 4.10: Preservation index for the Sterkfontein wood experiments, deposited one year outside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK105	Woody tissue	Outside cave	No	One year	2
STK106	Woody tissue	Outside cave	No	One year	2
STK29	Woody tissue	Outside cave	Yes	One year	2
STK30	Woody tissue	Outside cave	Yes	One year	3

Table 4.11: Preservation index for the Sterkfontein wood experiments, deposited four weeks inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK99	Woody tissue	In cave	No	Four weeks	5
STK100	Woody tissue	In cave	No	Four weeks	5
STK103	Woody tissue	In cave	Yes	Four weeks	5
STK104	Woody tissue	In cave	Yes	Four weeks	5

Table 4.12: Preservation index for the Sterkfontein wood experiments, deposited four weeks outside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK107	Woody tissue	Outside cave	No	Four weeks	2
STK108	Woody tissue	Outside cave	No	Four weeks	2
STK31	Woody tissue	Outside cave	Yes	Four weeks	3
STK32	Woody tissue	Outside cave	Yes	Four weeks	3-4

Wood is preserved well inside the cave at Sterkfontein. Interestigly, the unburied/exposed samples appear to preserve best. Both the four-week and one-year experiments show that preservation outside the cave is poor.

4.2.3 Muscle tissue sample

The results of the replicated and weathered muscle tissue sample are discussed in the following paragraphs. Tables 4.13-4.16 present the details of the experimental settings and the quantified preservation results.

STK113 and STK114: PI 3 and PI 3 (Figures B.77 and B.78)
The working edges are covered in hyphae and it is difficult to see the muscle tissue underneath. The tissue that is visible is birefringent. The residues are confined to the edges and many spores are present in the hyphae layer. For the intelligibility of the STK113 distribution maps, I did not indicate spores on it.

STK115 and STK116: PI 4 and PI 4 (Figures B.79 and B.80)
Along the working edges the muscle tissue is covered by the hyphae and as a result the residues are difficult to distinguish. Clear patches of muscle tissue occur in the interior of the tools. Some residue deposits are visible with the naked eye. Hyphae and spores are abundant.

STK117 and STK118: PI 3 and PI 3

STK117 probably broke and lost its cutting edge; as a result only one patch with muscle residue is present. Some hyphae and a few spores also occur. The working edge of STK118 is covered in hyphae and spores. Some muscle tissue is visible through the hyphae. The muscle tissue is birefringent.

STK119 and STK120: PI 3 and PI 3

Extensive hyphae are present all over the tool surface, but spores are absent. Along the working edges the hyphae deposits are extremely thick and the muscle tissue is hardly visible. Muscle tissue is concentrated along the working edges, but on STK119 on the dorsal side a tissue and hyphae spot is present in the middle of the tool. This could be the result of handling during the experiments.

STK121 and STK122: PI 1 and PI 1 (Figure B.79 and B.80)

On both tools, one side shows better residue preservation than on the other (see distributions maps). Occasional muscle tissue deposits occur and relatively more fat is present than on the previously discussed tools with muscle tissue residues. Many hyphae and spores are present, which are associated with muscle tissue, but also with sediment organics.

STK123 and STK124: PI 4 and PI 4

Along the working edge thick muscle tissue deposits are present. Some fatty bone and fat is also present. A few of these animal residues are invasive. Some spores and hyphae are present.

STK45 and STK46: PI 1 and PI 0

STK45: One muscle tissue deposit and one possible tissue deposit are present on the ventral side of this tool.

STK46: This tool contains no muscle tissue remains.

Hyphae are abundant on both tools.

STK47 and STK48: PI 2 and PI 1 (Figures B.83 and B84)

Both tools preserve few muscle tissue deposits. The deposits are covered in hyphae and hyphae interfere with the analysis. Spores are also abundant. Two hairs are present on STK47.

Table 4.13: Preservation index for the Sterkfontein muscle tissue experiments, deposited one year inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK113	Muscle tissue	In cave	No	One year	3
STK114	Muscle tissue	In cave	No	One year	3
STK117	Muscle tissue	In cave	Yes	One year	3
STK118	Muscle tissue	In cave	Yes	One year	3

Table 4.14: Preservation index for the Sterkfontein muscle tissue experiments, deposited one year outside the cave

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK121	Muscle tissue	Outside cave	No	One year	1
STK122	Muscle tissue	Outside cave	No	One year	1
STK45	Muscle tissue	Outside cave	Yes	One year	1
STK46	Muscle tissue	Outside cave	Yes	One year	0

Table 4.15: Preservation index for the Sterkfontein muscle tissue experiments, deposited four weeks inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK115	Muscle tissue	In cave	No	Four weeks	4
STK116	Muscle tissue	In cave	No	Four weeks	4
STK119	Muscle tissue	In cave	Yes	Four weeks	3
STK120	Muscle tissue	In cave	Yes	Four weeks	3

Table 4.16: Preservation index for the Sterkfontein muscle tissue experiments, deposited four weeks outside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK123	Muscle tissue	Outside cave	No	Four weeks	4
STK124	Muscle tissue	Outside cave	No	Four weeks	4
STK47	Muscle tissue	Outside cave	Yes	Four weeks	2
STK48	Muscle tissue	Outside cave	Yes	Four weeks	1

After four weeks there are no major changes in residue quantities, although, it appears that the buried samples are more deteriorated. After one year the differences between deposition in- and outside the cave become apparent. The preservation inside is good, but outside it is poor.

4.2.4 Bone sample

The results of the replicated and weathered bone sample are discussed in the following paragraphs. Tables 4.17-4.20 present the details of the experimental settings and the quantified preservation results.

STK129 and STK130: PI 4
STK129 was not recovered.
On STK130 fatty bone and fat residues are restricted to the working edge and most residues are located on the ventral side. The residues on the edge are continuous and unambiguous. Hyphae and spores are absent.

STK131 and STK 132: PI 4 and PI 4
Clear bone and fat deposits are present along the working edges, although part of the working edge of STK131 probably broke off. Some areas have extensive hyphae deposits. Spores are absent.

STK133 and STK 134: PI 2 and PI 2 (Figures B.85 and B.86)
A few bone and fat deposits are present along the edges. Hyphae occur on STK134, but not STK133. Hyphae are not associated with the residues. Spores are absent on both tools.

STK135 and STK136: PI 4 and PI 4 (Figures B.87 and B.88)
Clear fatty bone residues occur along the working edges. On STK135 some fatty bone is present towards the centre of the tool, which probably results from handling. On STK135 extensive hyphae and spores are present in association with the fatty bone and fat remains. On STK136 few hyphae and spores are present.

STK137 and STK138: PI 1 and PI 1 (Figures B.87 and B.88)
With only occasional bone and fat residues present, it is difficult to reconstruct the working edges. STK138 contains slightly more fatty bone residues than STK137 and the ventral side of STK138 contains many contaminants. Some hyphae and spores are present on the remaining surfaces. Only one fatty bone deposit is preserved on the ventral side of.

STK139 and STK140: PI 3-2 and PI 3-2

A few residues spots are present on the working edges. Only the ventral side of STK140 shows a small area with continuous residues. Hyphae and spores are present, but they are not as abundant as in STK 135.

STK61 and STK62: PI 2

STK61 was not recovered.

STK62 contains several fatty bone deposits. The residues are strictly confined to the edges. Some hyphae are present as are several sediment deposits, which contain charcoal and pollen.

STK63 and STK64: PI 3 and PI 3 (Figures B.91 and B.92)

These tools contain several bone deposits along the edges. In addition fat is relatively abundant on both artefacts. On STK63, thick deposits of hyphae and spores are present.

Table 4.17: Preservation index for the Sterkfontein bone experiments, deposited one year inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK129	NOT RECOVERED				
STK130	Bone	In cave	No	One year	4
STK133	Bone	In cave	Yes	One year	2
STK134	Bone	In cave	Yes	One year	2

Table 4.18: Preservation index for the Sterkfontein bone experiments, deposited one year outside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK137	Bone	Outside cave	No	One year	1
STK138	Bone	Outside cave	No	One year	1
STK61	NOT RECOVERED				
STK62	Bone	Outside cave	Yes	One year	2

Table 4.19: Preservation index for the Sterkfontein bone experiments, deposited four weeks inside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK131	Bone	In cave	No	Four weeks	4
STK132	Bone	In cave	No	Four weeks	4
STK135	Bone	In cave	Yes	Four weeks	4
STK136	Bone	In cave	Yes	Four weeks	4

Table 4.20: Preservation index for the Sterkfontein bone experiments, deposited four weeks outside the cave.

Experiment number	Residue type	Situation	Buried	Time	Preservation index
STK139	Bone	Outside cave	No	Four weeks	3-2
STK140	Bone	Outside cave	No	Four weeks	3-2
STK63	Bone	Outside cave	Yes	Four weeks	3
STK64	Bone	Outside cave	Yes	Four weeks	3

Bone residues are preserved at Sterkfontein, but the short experiments preserve them best. After one year few residues remain on the tools outside the cave. Inside the preservation might be better and it is unfortunate that STK129 was lost. The buried samples from inside the cave appear to have deteriorated more.

4.2.5 Discussion

After four weeks of deposition the residues are still present in large quantities on the tools. Hyphae grow abundantly on samples from both in and outside the cave. The hyphae did not develop spores within four weeks. Most sets of experiments show that the exposed experiments preserve better and this is in contrast with the Sibudu results, where the exposed sample deteriorated slightly more than the buried sample. The moist sediment outside Sterkfontein contains more micro-organisms, such as earthworms, than the Sibudu sediment, leading to higher oxygen levels and a better permeability at Sterkfontein. This leads to higher bioactivity, for example more fungus and bacteria, and poor residue preservation. In addition, the dry air in the Sterkfontein cave dries the residues and perhaps inhibits breakdown.

Some residue types preserve better than others. Bone and plant tissue preserve well, but muscle tissue and starch appear more vulnerable. Preservation is poor outside the cave; inside the residues are sheltered from rain and wind. The sediment inside the cave is also poorer in humus than outside the cave and there probably is less bioactivity inside than outside the cave.

The one-year results are more variable. Figures 4.1, 4.2, 4.3 and 4.5 summarise the Sterkfontein one-year results. Figure 4.1 presents the results for the exposed samples. Figure 4.2 illustrates the results for the buried samples. Figure 4.3 presents the results for the sample deposited inside the cave and Figure 4.4 presents the results from outside the cave. These figures illustrate that preservation inside the cave is better

than outside. Inside the cave, the buried samples contained more residues that the exposed samples. Outside the cave the exposed sample preserves slightly more residues, but with muscle tissue the buried sample scores higher. Interestingly, the tool side, which faced up and was exposed to the elements, preserved the least residues. The side, which faced down preserved the most residues. It is unclear why the buried sample does not score better than the exposed sample.

Starch grains deteriorate quickly both in- and outside the cave; they are extremely vulnerable. Muscle tissue preserves slightly better, but preserves poorly outside the cave. Spores are present mainly on the starch and muscle tissue samples, indicating rapid fungus growth on the residue types. Muscle tissue probably scores well in Figure 4.4 because hyphae obscured the analysis. This means that I was not certain how much muscle tissue was present under the hyphae layer and therefore assigned an index that was probably slightly too high. Bone and plant tissue generally preserve better than the other residue types, but once buried they are also more vulnerable.

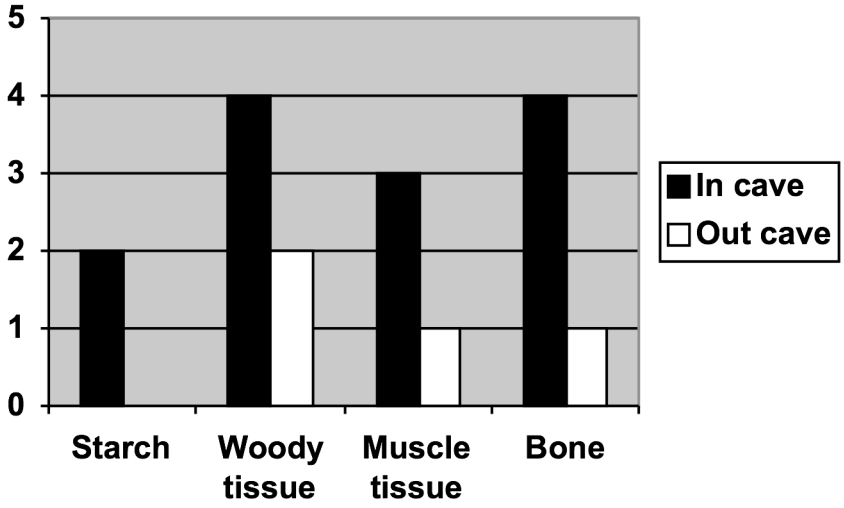


Figure 4.1: Preservation index for the Sterkfontein samples that were exposed for one year.

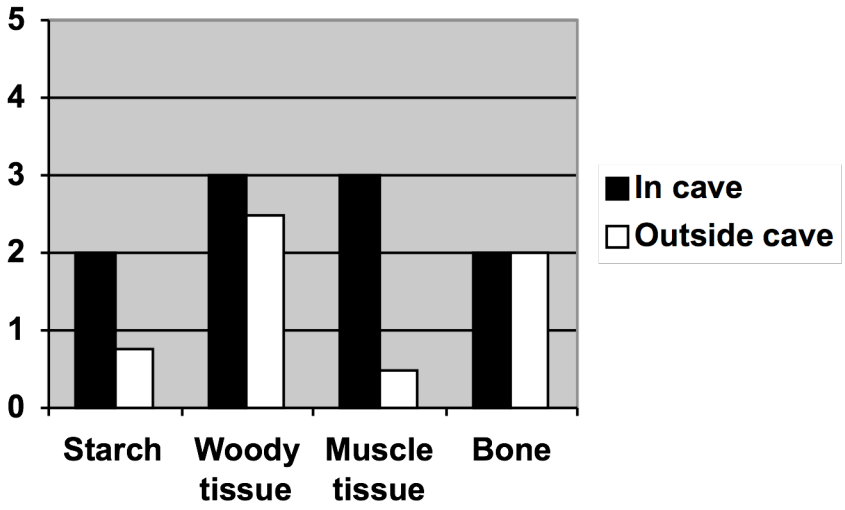


Figure 4.2: Preservation index for the Sterkfontein samples that were buried for one year.

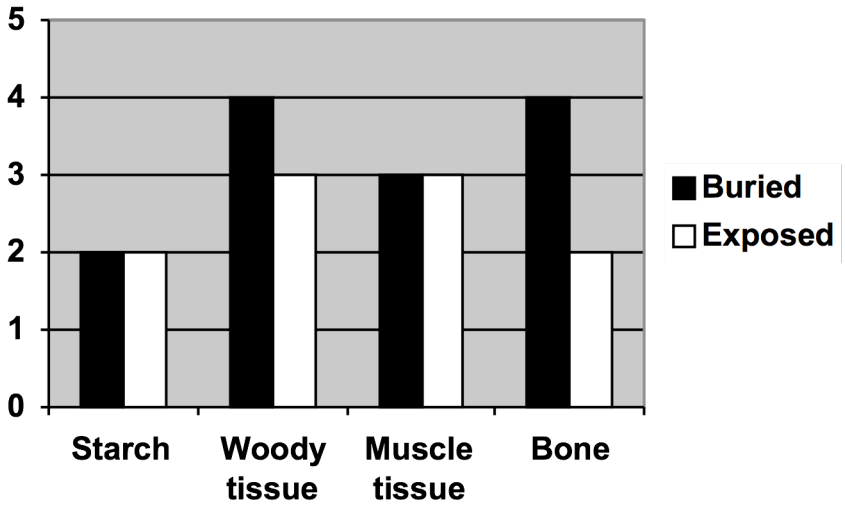


Figure 4.3: Preservation index for the Sterkfontein samples that were deposited inside the cave for one year.

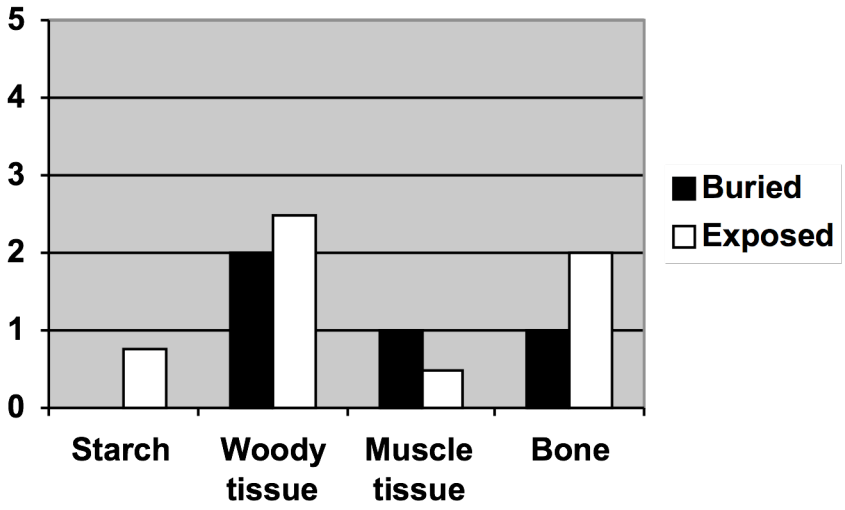


Figure 4.4: Preservation index for the Sterkfontein samples that were deposited outside the cave for one year.

4.3 REPLICATED TOOLS FROM SIBUDU

The variables tested at Sibudu are: residue type, time and the influence on residues of surface deposition in a shelter as well as the influence on residues of burial in a shelter deposit. Ten sets of experimental tools were set out in and outside the cave (Figure 2.3); some tools were buried and others were left on the sediment surface. Some experimental tools were collected after four weeks and others after 12 months. The experimental residues are: starch, woody plant tissue, muscle tissue and bone. Every set of experimental tools had a blank control sample that helped me to understand the hornfels and the sediment contaminants. I discuss the results per residue type and at the end of every section I summarise the results.

4.3.1 Starch sample

The results of the replicated and weathered starch sample are discussed in the following paragraphs. Tables 4.21-4.24 present the details of the experimental settings and the quantified preservation results.

SB81 and SB82: PI 4 and PI 4

Thick deposits of starch grains are present along the edges and more in the interior of the flake. Single grains and small clusters dominate towards the centre of the tool. Hyphae are present all over the tool's surface, but they do not obscure the view of residues. No spores occur.

SB83 and SB84: PI 4 and PI 4

Thick starch deposits are present along the edges. Towards the centre of the tool less starch occurs and often in small clusters and single grains. On SB83 several large deposits with epidermal tissue are present. Some hyphae, but no spores are present.

SB85 and SB86: PI 3 and PI 3 (Figures B.29 and B.30)

The working edges are covered in hyphae and spores and in some areas starch grains can be observed through a hyphae blanket. Only small amounts of starch are preserved. The starch grains are opaque due to contact with hyphae. Epidermal tissue is also present.

SB87 and SB88: PI 4 and PI 4 (Figures B.31 and B.32)

Clear starch residue deposits are present. Thicker deposits are present along the edge and small clusters and single grains occur in the interior of the flake. Epidermal tissue is also present. No hyphae or spores are visible. Note that the ventral side of SB88 contains sediment.

SB89 and SB90: PI 0 and PI 0

No starch is present on these tools. Epidermal tissue is also absent

SB91 and SB92: PI 5 and PI 5 (Figures B.33 and B.34)

Thick starch deposits occur all over the surface of the tools. Because the starch deposits are so thick and appear completely undamaged, these tools are given a PI of 5. Some pieces of epidermal tissue are present. Few hyphae occur, but no spores.

SB93 and SB94: PI 0 and PI 0 (Figure B.35)

No starch is preserved on either of the tools. One piece of epidermal tissue is present on SB93 (Figure B.36). No hyphae or spores are present.

SB95 and SB96: PI 4 and PI 4

The tools are covered in starch grains. Along the edges thicker deposits occur and in the interiors only single grains are present. Extensive hyphae occur on some edges, but there are no spores. Many pieces of epidermal tissue are present.

Table 4.21: Preservation index for the Sibudu starch experiments, deposited for one year inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB81	Starch	In shelter	No	One year	4
SB82	Starch	In shelter	No	One year	4
SB85	Starch	In shelter	Yes	One year	3
SB86	Starch	In shelter	Yes	One year	3

Table 4.22: Preservation index for the Sibudu starch experiments, deposited for one year outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB89	Starch	Drip line	No	One year	0
SB90	Starch	Drip line	No	One year	0
SB93	Starch	Drip line	Yes	One year	0
SB94	Starch	Drip line	Yes	One year	0

Table 4.23: Preservation index for the Sibudu starch experiments, deposited for four weeks inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB83	Starch	In shelter	No	Four weeks	4
SB84	Starch	In shelter	No	Four weeks	4
SB87	Starch	In shelter	Yes	Four weeks	4
SB88	Starch	In shelter	Yes	Four weeks	4

Table 4.24: Preservation index for the Sibudu starch experiments, deposited for four weeks outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB91	Starch	Drip line	No	Four weeks	5
SB92	Starch	Drip line	No	Four weeks	5
SB95	Starch	Drip line	Yes	Four weeks	4
SB96	Starch	Drip line	Yes	Four weeks	4

Starch residues appear to preserve quite well inside Sibudu Cave. No starch grains were preserved on the tools deposited for one year at the drip line. The one-year samples from inside the cave did not preserve as well as the short experiments. Exposed samples preserve slightly better than buried samples.

4.3.2 Wood tissue sample

The results of the replicated and weathered wood tissue sample are discussed in the following paragraphs. Tables 4.25-4.28 present the details of the experimental settings and the quantified preservation results.

SB97 and SB98: PI 3 and PI 3

Along working edges macerated plant tissue and fibres are present. The deposits are not thick, but continuous along parts of working edges. The woody tissue residues are restricted to the edges. Several resin spots with small starch grains occur. Some hyphae are present, but spores do not occur.

SB99 and SB100: PI 4 and PI 4 (Figures B.37 and B.38)

Abundant wood residues occur along the working edges of both tools. Some resin spots are present. Associated with the wood tissue are small starch grains. Hyphae are present, but spores are not.

SB101 and SB102: PI 3 and PI 3

One side of both the tools contains more residues than the other, for example, the ventral side of SB101 contains fewer residues than the dorsal side (see Figures B.39 and B.40). The working edges can easily be distinguished and contain wood tissue, fibres and resin. The residue deposits are not thick and invasive. No hyphae or spores are present.

SB103 and SB104: PI 4 and PI 4

Macerated wood tissue is abundant on these tools and plant fibres and resin are also present, but the residues are confined to the working edges. A few hyphae occur and no spores are present.

SB105 and SB106: PI 2 and PI 2 (Figures B.41 and B.42)

The tools only contain a few residues. Macerated wood tissue and fibres are restricted to the edges. No hyphae or spores are present. Note the abundant insect remains on SB106.

SB 107 and SB108: PI 4 and PI 4

Wood residues are abundant and occur almost continuously along the working edges of these tools. The plant residues are restricted to the edge. Hyphae and spores are absent.

SB109 and SB110: PI 3 and PI 3

One side of these tools contains fewer residues than the other side, which is similar to the unequal preservation on SB101 and SB102. Thin but continuous plant tissue deposits and fibres occur along the working edges. Hyphae and spores are absent.

SB111 and SB112: PI 4 and PI 4 (Figures B.43 and B.44)

The residues are preserved in thick and continuous patches along the working edges. On SB111 the plant tissue deposits are slightly invasive on the dorsal side (Figure B.44). No hyphae or spores are present.

Table 4.25: Preservation index for the Sibudu wood experiments, deposited for one year inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB97	Woody tissue	In shelter	No	One year	3
SB98	Woody tissue	In shelter	No	One year	3
SB101	Woody tissue	In shelter	Yes	One year	3
SB102	Woody tissue	In shelter	Yes	One year	3

Table.4.26: Preservation index for the Sibudu wood experiments, deposited for one year outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB105	Woody tissue	Drip line	No	One year	2
SB106	Woody tissue	Drip line	No	One year	2
SB109	Woody tissue	Drip line	Yes	One year	3
SB110	Woody tissue	Drip line	Yes	One year	3

Table 4.27: Preservation index for the Sibudu wood experiments, deposited for four weeks inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB99	Woody tissue	In shelter	No	Four weeks	4
SB100	Woody tissue	In shelter	No	Four weeks	4
SB103	Woody tissue	In shelter	Yes	Four weeks	4
SB104	Woody tissue	In shelter	Yes	Four weeks	4

Table 4.28: Preservation index for the Sibudu wood experiments, deposited for four weeks outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB107	Woody tissue	Drip line	No	Four weeks	4
SB108	Woody tissue	Drip line	No	Four weeks	4
SB111	Woody tissue	Drip line	Yes	Four weeks	4
SB112	Woody tissue	Drip line	Yes	Four weeks	4

Woody tissue preserves well at Sibudu. The one-year experiments show more deterioration than the short four-week experiments. Perhaps the buried samples are slightly better preserved. The one-year samples deposited around the drip-line are least preserved.

4.3.3 Muscle tissue sample

The results of the replicated and weathered muscle tissue sample are discussed in the following paragraphs. Tables 4.29-4.32 present the details of the experimental settings and the quantified preservation results.

SB113 and SB114: PI 3-2 and PI 2 (Figures B.45 and B46)
There are some areas with possible muscle tissue residues and others with clear tissue present. Hyphae and spores are abundant and obscure the possible residues. The residues are mainly located along the working edges. Fat is also present.

SB115 and SB116: PI 5 and PI 5
Extremely rich and thick residues occur along the edges, and the deposits also reach far into the interior of the tool. The residues consist of muscle tissue, macerated muscle tissue, fat and some bone. Some deposits are visible with the naked eye. Some hyphae are present, but spores are absent.

SB117 and SB118: PI 2 and PI 2 (Figures B.47 and B.48)
The working edges can easily be reconstructed by hyphae, which are growing on top of the muscle tissue. The hyphae layers are extremely thick and visible with the naked eye. The muscle tissue, under the fungus, is deteriorated. Some fatty spots are present. No spores occur.

SB119 and SB120: PI 4-3 and PI 4-3

Small and occasional deposits of muscle tissue and fat occur along the cutting edge and a few deposits are located away from the edges. Hyphae and spores are absent.

SB121 and SB122: PI 0 and PI 0

Residues are absent.

SB123 and SB124: PI 4-3 and PI 4-3 (Figures B.49 and B.50)

One side of the tools contains more residues than the other. There is less fatty bone present on SB1123 and SB124 than on the previously discussed tools. Clear muscle tissue deposits occur along the working edges, some cases the deposits are thick and in others the deposits are thinner and occasional. Hyphae are present, but spores are absent.

SB125 and SB126: PI 1 and PI 1 (Figures B.51 and B.52)

Occasional residues occur around the working edges. There are many ambiguous residues, which are probably altered by hyphae or are hyphae. Hyphae are present in a gelatinous form, probably forming mycelium. By this I mean that it is not possible to distinguish clear strands, but that the hyphae are compressed together, creating a gelatinous mass from which short strands emerge (Figures B.130 and B.131).

SB127 and SB128: PI 5 and PI 5

The tools are covered in thick and continuous deposits of muscle tissue, which can be seen with the naked eye. The muscle tissue is thickest along the working edges, but also occurs towards the centre of the tools. Almost no fat is present. Hyphae are present, but not excessively. Spores are absent.

Table 4.29: Preservation index for the Sibudu muscle tissue experiments, deposited one year inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB113	Muscle tissue	In shelter	No	One year	3-2
SB114	Muscle tissue	In shelter	No	One year	2
SB117	Muscle tissue	In shelter	Yes	One year	2
SB118	Muscle tissue	In shelter	Yes	One year	2

Table 4.30: Preservation index for the Sibudu muscle tissue experiments, deposited one year outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB121	Muscle tissue	Drip line	No	One year	0
SB122	Muscle tissue	Drip line	No	One year	0
SB125	Muscle tissue	Drip line	Yes	One year	1
SB126	Muscle tissue	Drip line	Yes	One year	1

Table 4.31: Preservation index for the Sibudu muscle tissue experiments, deposited four weeks inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB115	Muscle tissue	In shelter	No	Four weeks	5
SB116	Muscle tissue	In shelter	No	Four weeks	5
SB119	Muscle tissue	In shelter	Yes	Four weeks	4-3
SB120	Muscle tissue	In shelter	Yes	Four weeks	4-3

Table 4.32: Preservation index for the Sibudu muscle tissue experiments, deposited four weeks outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB123	Muscle tissue	Drip line	No	Four weeks	4-3
SB124	Muscle tissue	Drip line	No	Four weeks	4-3
SB127	Muscle tissue	Drip line	Yes	Four weeks	5
SB128	Muscle tissue	Drip line	Yes	Four weeks	5

Muscle tissue is better preserved in the shelter than outside, but after one year few residues remain. The buried samples show slightly better preservation than the exposed ones.

4.3.4 Bone sample

The results of the replicated and weathered bone sample are discussed in the following paragraphs. Tables 4.33-4.36 present the details of the experimental settings and the quantified preservation results.

SB129 and SB130: PI 4 and PI 3 (Figures B.53 and B.54)

SB129: Clear residues are preserved, but they are confined to the working edge. Bone is exceptionally fatty and fat residues are also abundant. White fungus fruiting bodies are present and hyphae are present all over the bone and fat deposits.

SB130: Only a few bone residues are present on the edges. The residues are very fatty in appearance. Hyphae are present.

SB131 and SB132: PI 3 and PI 3 (Figures B.55 and B.56)
Some bone residues are present and they are all extremely fatty. Fat is also present on SB131 and SB132. Because bone and fat are concentrated along the edges, the working edge is easily distinguished. White fungus fruiting bodies and hyphae are present on SB132. The bone, fat and muscle tissue residues in Figure B.56 illustrate the association of residues.

SB133 and SB134
Not recovered.

SB135 and SB136: PI 4 and PI 4
Evident fatty bone and fat deposits occur along the working edges. No residues occur in the centre of the tools. Some residue deposits are reasonably thick, but most are thin. White fungus fruiting bodies and hyphae are present.

SB137 and SB138: PI 0 and PI 0
Residues are absent.

SB139 and SB140: PI 4 and PI 4 (Figures B.57 and B.58)
Fatty bone residues are continuously present along the working edge. The bone is extremely fatty. In contrast to the previously discussed tools, white fungus fruiting bodies or hyphae are not present nor are any black spores. Note that the bone residues are confined to the edges.

SB141 and SB142: PI 2 and PI 1 (Figures B.59 and B.60)
A few fatty bone and fat residues are present. The residues are confined to the working edge only. There are some hyphae present, but no spores.

SB143 and SB144: PI 4 and PI 4
Clear fatty bone residues are preserved along the edges. Some deposits are reasonably thick. SB143 contained some connective fibres. White fungus fruiting bodies and hyphae are absent.

Table 4.33: Preservation index for the Sibudu bone experiments, deposited one year inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB129	Bone	In shelter	No	One year	4
SB130	Bone	In shelter	No	One year	3
SB133	NOT RECOVERED				
SB134	NOT RECOVERED				

Table 4.34: Preservation index for the Sibudu bone experiments, deposited one year outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB137	Bone	Drip line	No	One year	0
SB138	Bone	Drip line	No	One year	0
SB141	Bone	Drip line	Yes	One year	2
SB142	Bone	Drip line	Yes	One year	1

Table 4.35: Preservation index for the Sibudu bone experiments, deposited four weeks inside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB131	Bone	In shelter	No	Four weeks	3
SB132	Bone	In shelter	No	Four weeks	3
SB135	Bone	In shelter	Yes	Four weeks	4
SB136	Bone	In shelter	Yes	Four weeks	4

Table 4.36: Preservation index for the Sibudu bone experiments, deposited four weeks outside the shelter.

Experiment number	Residue type	Situation	Buried	Time	Preservation Index
SB139	Bone	Drip line	No	Four weeks	4
SB140	Bone	Drip line	No	Four weeks	4
SB143	Bone	Drip line	Yes	Four weeks	4
SB144	Bone	Drip line	Yes	Four weeks	4

The one-year samples clearly show that bone is better preserved inside the cave than outside. The buried samples preserve better than the exposed ones.

4.3.5 Discussion

After four weeks of deposition, all residues types were abundant in all settings. Hyphae grow on these samples, but have not developed fruiting bodies or spores yet. As the residues are abundant, the contaminants are easily distinguished. Most experimental sets show that buried residues preserve best. Perhaps this is due to a lack of oxygen, airborne fungi/spores or moisture from the air. Some residue types preserve better than others. Bone and woody tissue preserve well, but muscle tissue and starch are vulnerable.

There are more interesting results for the one-year experiments. Inside the shelter preservation is good, residues are abundant and there is no clear distinction between buried and exposed samples. The lack of distinction is influenced by the missing buried bone sample. At this stage it appears that at least starch benefits from being exposed and preserves better than the buried counterpart. The experimental tools were deposited in April, which is at the onset of the dry season. As a result, the exposed samples inside the cave dried quickly; ensuring preservation. The residues at the drip line, however, deteriorated much more and in some cases no residues are preserved. For this set, the buried samples show better preservation. Although the exposed samples at the dripline also dried in the dry season, they rehydrated when the wet season came. Starch and muscle tissue do not preserve well, whereas bone does. Woody tissue preserves much better than the other types.

Figures 4.5 and 4.6 illustrate the one-year experiments. Figure 4.5 shows the exposed samples and from Tables 4,7 and 4,8 it is clear that inside the cave preservation is best and that woody tissue preserves best. Figure 4.5 shows that the residues on the buried samples preserve slightly less well than the residues on the exposed tools. Starch and muscle tissue are fragile.

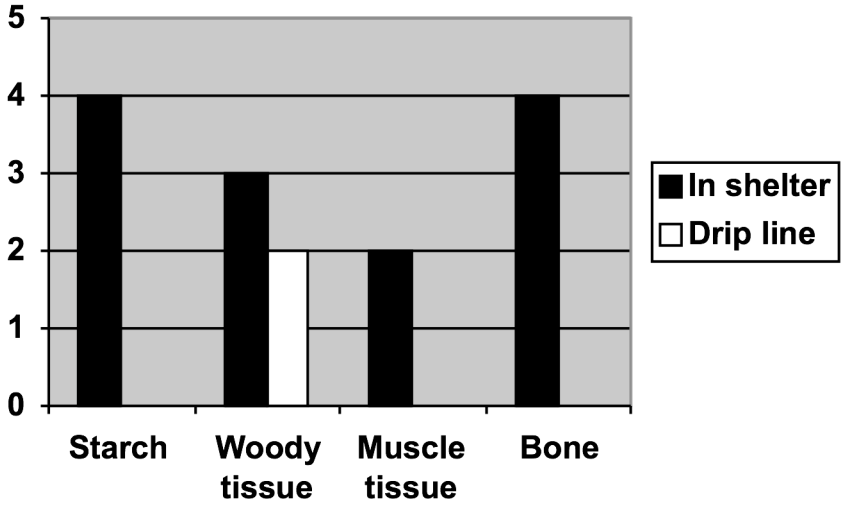


Figure 4.5: Preservation index for the Sibudu samples that were exposed for one year.

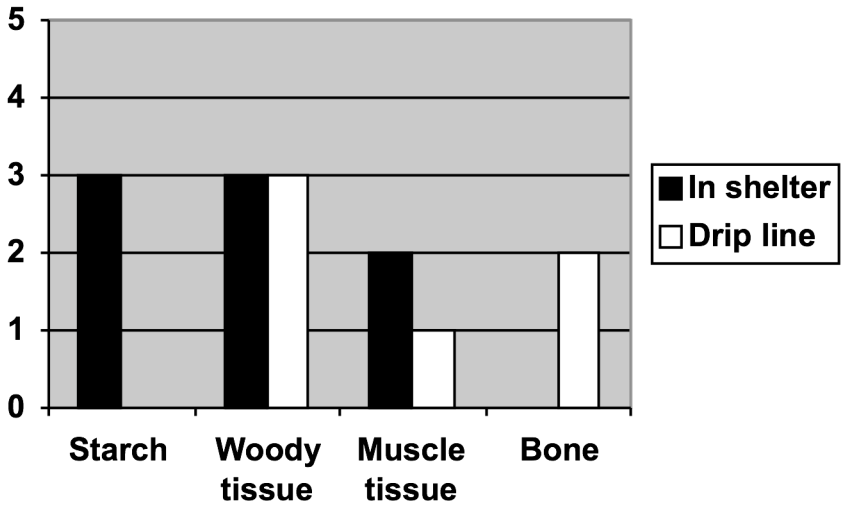


Figure 4.6: Preservation index for the Sibudu samples that were buried for one year. Note that the bone sample, inside the shelter was not recovered.

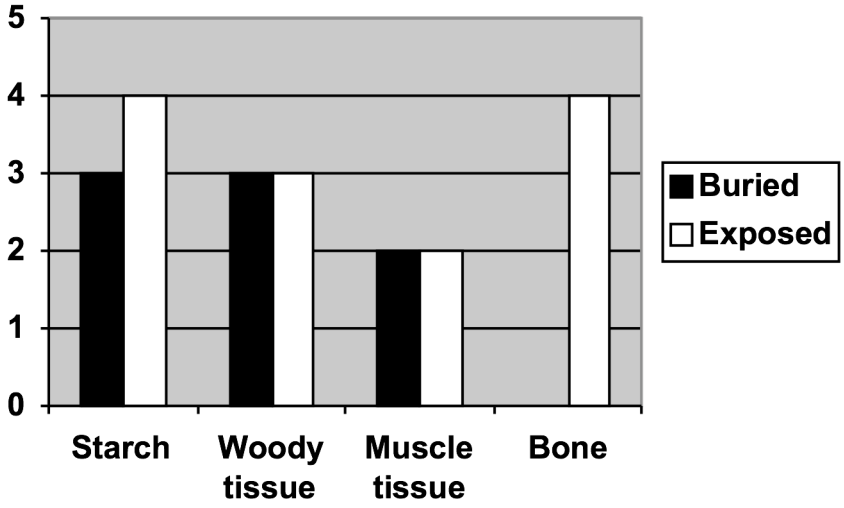


Figure 4.7: Preservation index for the Sibudu samples that were deposited inside the shelter for one year. Note that the buried bone sample was not recovered.

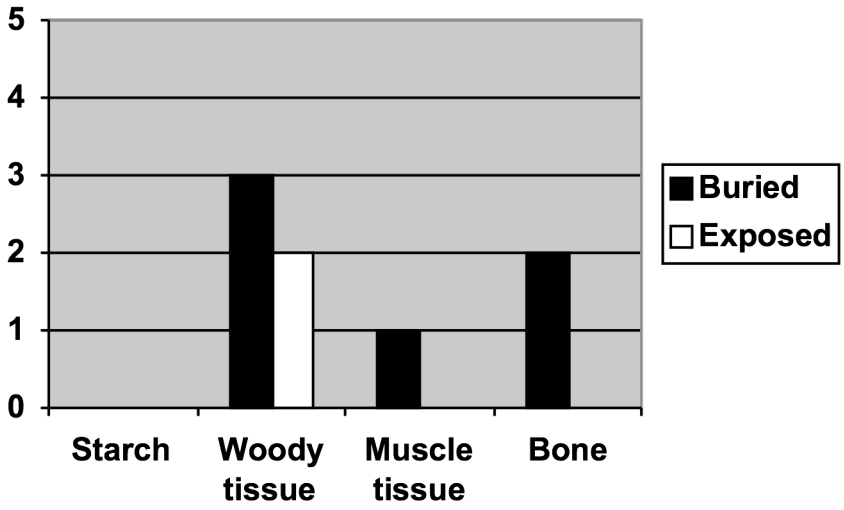


Figure 4.8: Preservation index for the Sibudu samples that were deposited outside the shelter for one year.

4.4 WILLEMINA POLDER

In this part of the study I deposited 25 replicated flint tools in clay sediment at Willemina Polder in the South West of the Netherlands. Thirteen tools were buried and 12 were left exposed on the sediment surface. The tools were used on marrow fat/bone and starch to create corresponding residues and five blank tools were also included. An additional set of microscopes slides with sandpaper attached to them was also set out. On these I deposited blood, marrow/bone and starch residues. This set consisted of 88 samples including 16 blanks. Half of the sandpaper samples were deposited with residues facing down into the sediment and the other half facing up. All samples were left in their positions for six months. Most of the tools lost their proveniences as the tool numbers eroded off. The nine tools that were excavated without a number were all treated as used tools. However, it is possible that some of these tools belong to the blank sample and that tools with no use-related residues and thus an index score of zero are over-represented. I randomly renumbered the tools without a tool number from NLCX1 to NLCX9.

The sandpaper sample was completely lost. The field where the samples were deposited was ploughed and I only found broken microscope glasses, which often did not contain a number or a sandpaper patch.

NLC1: PI 0

No residues are present.

NLC17: PI 2-1 (Figures B.93 and B.94)

Few fatty bone and fat deposits are present on the ventral side; on the dorsal side no residues are preserved. The deposits are small and located on the very edge of the tool. There are no hyphae or spores present.

NLC19: PI 1

Occasional fat and muscle tissue deposits occur. The deposits are small and ambiguous. Some hyphae are present, but spores are absent.

NLC20: PI 1

Only one unambiguous fat residue is present on this tool. Some hyphae are present, but spores are absent.

NLCX1: PI 0

Residues are absent.

NLCX2: PI 0

Two pieces of epidermal tissue reveal that this must have been a starch sample. The starch grains, however, are not preserved. Some hyphae are present, but spores are absent.

NLCX3: PI 0

The tool label still contained a number eight and therefore it could either be a starch or a marrow/bone tool, but definitely not a blank. However, no residues are preserved.

NLCX4: PI 1 (Figures B.95 and B.96)

Occasional small bone flakes occur along the working edges of this tool. The fatty bone deposits around the platform are probably contaminants; they are extremely small and their location on the obtuse edge does not make sense. Hyphae and spores are absent.

NLCX5: PI 0

The tool label still contained the number six and therefore it could either be a starch or a marrow/bone tool, but definitely not a blank. Because NLCX7 was identified as bone/marrow tool, NLCX5 was a starch tool. However, no residues are preserved.

NLCX6: PI 0

Not one of the remains on this tool is unambiguous and although there are some possible fat and bone residues they do not follow the distinct bone pattern. They are not confined to the working edge and it is not clear which edge was the working edge. Therefore, I cannot identify any use-related residues on this tool.

NLCX7: PI 1

The tool label contained the number six and therefore it could either be a starch or a marrow/bone tool, but definitely not a blank. One bone flake is present on the edge and the tool is identified as a marrow/bone sample. No hyphae or spores are present.

NLCX8: PI 2-3 (Figures B.97 and B.98)

This sample was collected from the sediment surface. Three large and thick fatty bone and fat deposits are present along the distal edge on the ventral side. The stone under the bone and fat residues is abraded. The dorsal side contains no residues. Hyphae and spores are absent.

NLCX9: PI 0 (Figures B.99 and B.100)

This sample was collected from the surface and it preserved plant fibres and characteristic epidermal tissue, which can be seen with the naked eye. The epidermal tissue indicates that the tool was used on starch, but the starch grains are not present. Hyphae and spores are absent.

Table 4.37: Preservation index for the Willemina Polder experiments with proximity data.

Tool number	Residue type	Situation	Preservation index
NLC10	Starch	Buried	0
NLC17	Marrow/bone	Buried	2
NLC19	Marrow/bone	Buried	1
NLC20	Marrow/bone	Buried	1

Table 4.38: Preservation index for the Willemina Polder experiments without proximity data.

Tool number	Residue type	Reconstructed residue type	Situation	Preservation index
NLCX1	Unknown	Unknown	Buried	0
NLCX2	Unknown	Starch	Buried	0
NLCX3…8	Unknown	Starch	Buried	0
NLCX4	Unknown	Marrow/bone	Buried	2
NLCX5…6	Unknown	Starch or marrow	Buried	0
NLCX6	Unknown	Unknown	Buried	0
NLCX7…6	Unknown	Marrow/bone	Buried	1
NLCX8	Unknown	Marrow/bone	Exposed	2-3
NLCX9	Unknown	Starch	Exposed	0

The clay sediment at Willemina Polder contains few contaminants and the sediment remains did not interfere greatly with the analysis. However, the clay sediment itself appears fatty under the microscope and this is problematic when viewing bone and fat residues.

On the starch tools no starch grains were preserved. Epidermal tissue, however, was often still present. For example, on NLCX9 no starch grains are present, but the tool did contain large deposits of epidermal tissue and fibres.

Marrow/bone preserve better than starch, but these tools never score high in the preservation index either. The buried samples performed considerably less well than the exposed ones. The tools without numbers that scored 0 are most likely either blanks or starch tools.

4.5 ZELHEM

In this part of the study I deposited 25 flint tools at Zelhem. 13 tools were buried and 12 were left exposed on the sediment surface. The tools were used on marrow fat/bone and starch to create corresponding residues and five blank tools were also included. Additionally a set of microscopes slides with sandpaper attached to them on which I deposited blood, fat/marrow and starch residues was also set out. The slides vary in grain size from small to large. In addition, I also set out a sample of glass slides, without sandpaper. This set comprised 88 samples including 16 blanks. Half of the slides were deposited with residues facing down into the sediment and the other half facing up, towards the humus layers. All samples were left to weather for six months.

4.5.1 Tools

NLS1 and NLS2 were not recovered.

NLS3-NLS5: PI 2, PI 3 and PI 2

Figures B.101 and B.102 present the distribution maps for NLS4.

Starch is present on the remaining tools from this exposed sample, however, not in large and thick deposits, but in thin, spread out small clusters. Many epidermal tissue deposits are present on the tools in this sample. Most starch deposits are affected by hyphae and appear opaque. The used edge can be reconstructed on the tools.

NLS6-NLS10: PI 0, PI 0, PI 2, PI 1-2 and PI 3

Figures B.103 and B.104 present the distribution maps for NLS10.

On the buried sample NLS6-NLS10 less starch is present than on the exposed sample. Again starch grains are spread out over the surface and do not appear in thick layers. Hyphae are present in association with the opaque grains. Epidermal tissue is also present on most of the tools, but on NLS7 epidermal tissue and starch grains are absent. The used edge can be reconstructed on all tools with starch grains.

NLS11-NLS15: PI 2, PI 2, PI 2, PI 1 and PI 3

Figures B.105 and B.106 present the distribution maps for NLS14.

This sample with marrow/bone residues was exposed. Only a few residues are present on NLS11-NLS14. On NLS15 more fat and bone occurs and a working edge can be reconstructed. On all tools extremely fatty bone and bone flakes are present. Hyphae also occur.

NLS16-NLS20: PI 2, PI 3, PI 2, PI 0 and PI 2

Figures B.107 and B.108 present the distribution maps for NLS17

The results from the buried marrow/bone sample are similar to the results from the exposed sample. Only on NLS19 no residues are preserved. The remaining tools contain fat smears and fatty bone flakes. In addition, NLS16 contains one muscle tissue patch. Hyphae are present in larger deposits, but they are not always associated with the residues. NLS17 preserved slightly more residues than the other tools in this sample.

Table 4.39: Preservation index for the starch experiments in sandy sediment.

Tool number	Residue type	Situation	Preservation index
NLS1	NOT RECOVERED		
NLS2	NOT RECOVERED		
NLS3	Starch	Exposed	2
NLS4	Starch	Exposed	3
NLS5	Starch	Exposed	2
NLS6	Starch	Buried	0
NLS7	Starch	Buried	0
NLS8	Starch	Buried	2
NLS9	Starch	Buried	1-2
NLS10	Starch	Buried	3

Table 4.40: Preservation index for the bone/marrow experiments in sandy sediment at Zelhem.

Tool number	Residue type	Situation	Preservation index
NLS11	Bone/marrow	Exposed	2
NLS12	Bone/marrow	Exposed	2
NLS13	Bone/marrow	Exposed	2
NLS14	Bone/marrow	Exposed	1
NLS15	Bone/marrow	Exposed	3
NLS16	Bone/marrow	Buried	2
NLS17	Bone/marrow	Buried	3
NLS18	Bone/marrow	Buried	2
NLS19	Bone/marrow	Buried	0
NLS20	Bone/marrow	Buried	2

The exposed starch sample appears to have better preservation than the buried sample. However, two tools from the exposed sample were not recovered and this influences the conclusion. Epidermal tissue is abundant on the starch tools. There is no distinction between the preservation of marrow/bone in the buried or exposed samples. Preservation of fatty bone and fat is better than starch. In general, the preservation for both the starch and marrow/bone samples on sandy sediment is poor.

4.5.2 Sandpaper and glass slide experiments

The Preservation Index indicated after the tool numbers is the average score.

Blood

NLSBL1-24: PI 0

No blood residues are preserved. In a few cases, on the samples facing down, a deposit of spores is present in the middle of the sandpaper, located in the same area where the residues were deposited.

Table 4.41: Preservation index for the blood experiments on sandpaper. Up means that the residues faced upwards; Down means that the residue faced downwards.

Tool number	Residue type	Raw material	Situation	Preservation index
NLSBL1	Blood	Glass	Up	0
NLSBL2	Blood	Glass	Up	0
NLSBL3	Blood	Glass	Down	0
NLSBL4	Blood	Glass	Down	0
NLSBL5	Blood	Glass	Down	0
NLSBL6	Blood	Glass	Down	0
NLSBL7	Blood	Sandpaper small	Up	0
NLSBL8	Blood	Sandpaper small	Up	0
NLSBL9	Blood	Sandpaper small	Down	0
NLSBL10	Blood	Sandpaper small	Down	0
NLSBL11	Blood	Sandpaper small	Down	0
NLSBL12	Blood	Sandpaper small	Down	0
NLSBL13	Blood	Sandpaper medium	Up	0
NLSBL14	Blood	Sandpaper medium	Up	0
NLSBL15	Blood	Sandpaper medium	Down	0
NLSBL16	Blood	Sandpaper medium	Down	0
NLSBL17	Blood	Sandpaper medium	Down	0
NLSBL18	Blood	Sandpaper medium	Down	0
NLSBL19	Blood	Sandpaper large	Up	0
NLSBL20	Blood	Sandpaper large	Up	0
NLSBL21	Blood	Sandpaper large	Down	0
NLSBL22	Blood	Sandpaper large	Down	0
NLSBL23	Blood	Sandpaper large	Down	0
NLSBL24	Blood	Sandpaper large	Down	0

Starch

Glass

NLSS1-NLSS6: PI 1

On the samples facing up no residues are preserved, but the samples facing down have some starch preservation. The starch grains were often encapsulated in hyphae. No spores are present.

Sandpaper S

NLSS7-NLSS12: PI 0.5

On NLSS7 one piece of epidermal tissues is present, but starch grains are absent. On the other samples facing down only a few grains are present. The starch grains are present in small clusters and often as single grains associated with hyphae.

Sandpaper M

NLSS13-NLSS18: PI 0.5

NLSS14 contains one piece of epidermal tissue. The samples that faced down show some starch grains, but only in small clusters in association with hyphae. Because

they are damaged by hyphae, they are dull and opaque. This makes it difficult to distinguish starch grains from smaller quartz grains.

Sandpaper L

NLSS19-NLSS24: PI 0.4

NLSS20 contains a small piece of epidermal tissue and many spores. The samples facing down contain few starch grains. In comparison, far fewer grains occur on this coarse-grained sandpaper than on the small- and medium-grained sandpaper. The starch grains are partly absorbed by hyphae and take over viscous properties of the hyphae. The grains are therefore difficult to recognise.

Table 4.42: Preservation index for the starch experiments on sandpaper at Zelhem.

Tool number	Residue type	Raw material	Situation	Preservation index
NLSS1	Starch	Glass	Up	0
NLSS2	Starch	Glass	Up	0
NLSS3	Starch	Glass	Down	1
NLSS4	Starch	Glass	Down	1
NLSS5	Starch	Glass	Down	2
NLSS6	Starch	Glass	Down	2
NLSS7	Starch	Sandpaper small	Up	0
NLSS8	Starch	Sandpaper small	Up	0
NLSS9	Starch	Sandpaper small	Down	1
NLSS10	Starch	Sandpaper small	Down	0
NLSS11	Starch	Sandpaper small	Down	1
NLSS12	Starch	Sandpaper small	Down	1
NLSS13	Starch	Sandpaper medium	Up	0
NLSS14	Starch	Sandpaper medium	Up	0
NLSS15	Starch	Sandpaper medium	Down	1
NLSS16	Starch	Sandpaper medium	Down	1
NLSS17	Starch	Sandpaper medium	Down	0
NLSS18	Starch	Sandpaper medium	Down	1
NLSS19	Starch	Sandpaper large	Up	0
NLSS20	Starch	Sandpaper large	Up	0
NLSS21	Starch	Sandpaper large	Down	1
NLSS22	Starch	Sandpaper large	Down	1
NLSS23	Starch	Sandpaper large	Down	1-0
NLSS24	Starch	Sandpaper large	Down	0

No residues are preserved on the samples facing up. Regardless of the sandpaper the samples that face down generally show similar preservation. The glass samples preserve residues best. However, the glass samples were analysed under the direct light microscope and it was much easier to find the starch grains in this light. Perhaps, more starch grains are present on the sandpaper samples, but I was unable to identify them.

Fat/marrow

Most fat samples had a hard conglomeration of sediment attached to the surface. I think that the sediment absorbed the fat creating this hard layer.

Glass

NLSF1-NLSF6: PI 0

No residues are preserved on the glass samples. Hyphae and spore sheets are only present on the samples facing down.

Sandpaper S

NLSF7-NLSF12: PI 0.7

There are no residues present on the samples that faced up. The ones facing down, however, had a thick pack of sediment attached to the surface. When the sediment was removed a white layer appeared underneath it. This layer contained clear fat cells and hyphae strands. The area covered by this sediment and hyphae deposit was not as large as the original fat deposit.

Sandpaper M

NLSF13-NLSF18: PI 0.8

The two samples that faced up show no residue preservation. The ones facing down, however, had a thick pack of sediment attached to the surface and when it was removed, a white layer became visible. This layer was a mix of fat and hyphae strands. The hyphae dominated this deposit. The area covered by this fat and hyphae deposit was not as large as the original fat deposit. NLSF16 did not contain these white deposits, but only a sheet of dark hyphae and spores.

Sandpaper L: PI 0

NLSF19-NLSF24

No residues are preserved on these slides. However, on all the down-facing samples, a sheet of thick dark hyphae and spore is present.

Table 4.43: Preservation index for the fat experiments on sandpaper at Zelhem.

Tool number	Residue type	Raw material	Situation	Preservation index
NLSF1	Fat	Glass	Up	0
NLSS2	Fat	Glass	Up	0
NLSF3	Fat	Glass	Down	0
NLSF4	Fat	Glass	Down	0
NLSF5	Fat	Glass	Down	0
NLSF6	Fat	Glass	Down	0
NLSF7	Fat	Sandpaper small	Up	0
NLSF8	Fat	Sandpaper small	Up	0
NLSF9	Fat	Sandpaper small	Down	2
NLSF10	Fat	Sandpaper small	Down	2
NLSF11	Fat	Sandpaper small	Down	2
NLSF12	Fat	Sandpaper small	Down	2
NLSF13	Fat	Sandpaper medium	Up	0
NLSF14	Fat	Sandpaper medium	Up	0
NLSF15	Fat	Sandpaper medium	Down	1
NLSF16	Fat	Sandpaper medium	Down	0
NLSF17	Fat	Sandpaper medium	Down	2
NLSF18	Fat	Sandpaper medium	Down	2
NLSF19	Fat	Sandpaper large	Up	0
NLSF20	Fat	Sandpaper large	Up	0
NLSF21	Fat	Sandpaper large	Down	0
NLSF22	Fat	Sandpaper large	Down	0
NLSF23	Fat	Sandpaper large	Down	0
NLSF24	Fat	Sandpaper large	Down	0

Most striking from these results is the complete lack of preservation in the samples facing up. Figures 4.9 and 4.10 illustrate this point. It is striking that the small-grained sandpaper preserves the residues best for fat and that glass does not preserve any.

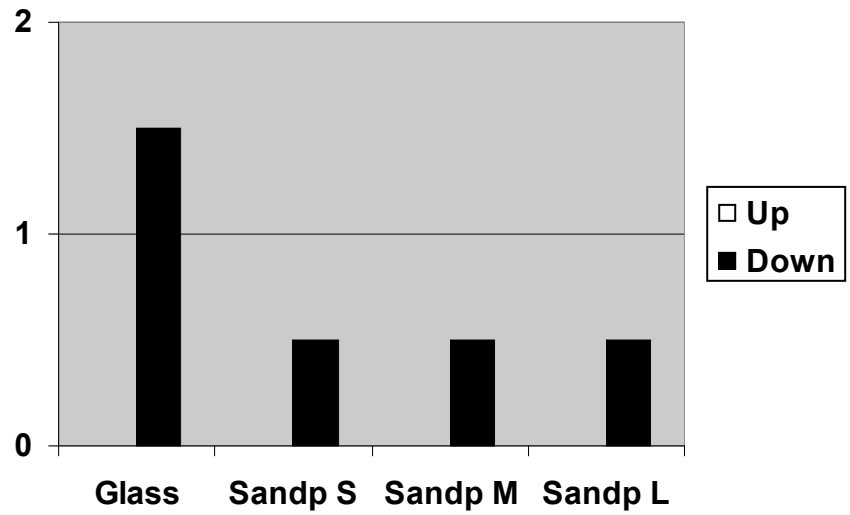


Figure 4.9: Average preservation index for starch experiments on glass and different types of sandpaper. No starch is preserved on the samples facing up.

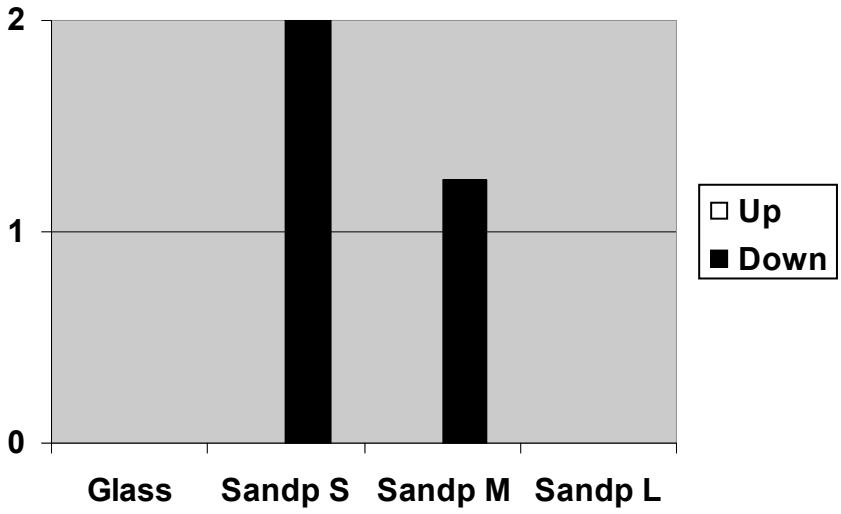


Figure 4.10: Average preservation index for fat experiments on glass and different types of sandpaper. No fat is preserved on the slides facing up.

4.5.3 Discussion

The sandy sediment used in these experiments does not preserve organic residues well. The samples facing down preserve residues better than the samples that faced upwards. The glass slide with sandpaper creates a protected environment, where residues preserve. The small-grained slides preserve residues best and the coarse-grained slides poorest. In the fat experiments, the glass slides with no sandpaper score as badly as the coarse-grained slides. In the starch experiments, however, the glass slides have the best score. A discrepancy from using two different types of microscopes caused the glass slide to score high. This example demonstrates how easily residues are overlooked and how carefully one needs to analyse samples.

Fat scored 0 on glass, but starch scored 1 on glass. Some residues need to attach to a surface in order to preserve. Fat probably partly soaks into the raw material of a stone tool or the sandpaper on a slide and the protecting sediment and hyphae also require an irregular surface to attach to. Glass-like surfaces are not irregular and therefore some types of residues do not preserve on these surfaces.

4.6 DISCUSSION

By studying blanks and sediment samples, and comparing these to genuine residues, it is possible to distinguish use-related residues from contaminants. The contextual approach, in which association and distribution of remains play an important role, helps greatly to identify use-related residues and post-depositional residues.

The distribution maps in Appendix B demonstrate that use-related residues have a meaningful distribution. In these experiments, residues are found along the working edges and sometimes on the obtuse ends and sides. Handling causes the latter. Different functions leave different distribution patterns and experimental research into hafting and tool use is important to understand residue patterns. Contaminants have a random distribution. In addition, the distribution maps in Appendix B also demonstrate that residues are associated. Starch grains are found in combination with epidermal tissue and fat is often found with bone and muscle tissue. Residue analysis must be conducted by using the contextual approach, as discussed in Chapter Two.

Quantification of residues is important; one plant fibre on a tool, for example, is not sufficient evidence for identifying a woodworking tool. From the experiments in this chapter it is evident that when residues are preserved they occur in larger quantities. By means of one fragmentary residue it is impossible to reconstruct tool function with distribution maps and because when there are no associated residues it is impossible to make a distinction between use-related and post-depositional residues.

Of all potential contaminants, I want to highlight starch. Starch is a challenging residue because it can so often be a contaminant. Sediments can contain large starch grains (>5 micrometre), but generally they contain larger quantities of small starch grains (2-5 micrometre). When hyphae break down the grains, they incorporate them within their gelatinous structure. First the starch grains are encapsulated by hyphae and later they are indistinguishable from the hyphae. This means that small starch grains seen on slides or tools are merely small starch grains and not partly deteriorated grains. Due to their size they probably are contaminants. If one also takes into account the vulnerability of starch, and keeps in mind how quickly starch disappears from a tool, it becomes clear that starch is an extremely problematic residue to interpret.

The experiments demonstrate that protective environments in caves and shelters, or on a smaller scale, under sandpaper-covered slides, have a positive effect on residue preservation. Sites with low bioactivity, such as Sibudu, have good residue preservation, whereas sites with high bioactivity, such as Willemina Polder have poor residue preservation.

In the following paragraphs I shall discuss the impact of the results that I have presented. In order to draw general conclusions that are valid across sites I divide the discussion according to topic.

4.6.1 Chain of events

The experiments help to get insight into the process of residue deterioration. The fat on the sandpaper experiments and the Sterkfontein starch experiments illustrate this process well. Figure B.109 presents fat as it looks prior to deposition. Fat globules are distinguishable, but after deposition, fat is compressed to a compact layer and the first hyphae appear (Figures B.110 and B.111). More hyphae grow on the fat deposit (Figure B.112), until they dominate the deposit (Figure B.113). Hyphae grow in strands (Figure B.114), and over time they form a complex web of strands and mycelia (Figure B.115). Finally the hyphae form a gelatinous deposit (Figures B.130 and B.131). The residues are absorbed by the fungus and gradually lose the characteristics that could be used for their identification: starch grains become dull and opaque and plant tissue starts resembling bone and muscle tissue (See paragraph 4.6.2). Gradually the fungi become more dominant and eventually no other residues are left. Just prior to this stage, the fungi start making and depositing spores (Figures B.116 to B.118). Due to lack of nutrition, the hyphae disappear. The tool's surface is now covered with spores (Figures B.119). The spores form a brown-black layer that in turn slowly decomposes over time (Figure B.120). Eventually only small clusters of spores or no spores remain.

Starch grains do not become smaller when they are absorbed by fungus. Hyphae encapsulate the grains and slowly absorb them into the gelatinous matrix until the grains disappear (Figures B.121 to B.124).

On the experimental tools I found hyphae and spores mainly on residues from starch and muscle tissue. The woody tissue and bone samples contain fewer spores and this is probably because their decomposition is more complex. On the starch and

muscle tissue residues, which break down easily, hyphae grow faster and deposit their spores more quickly than on more resilient residues.

Hyphae appear on blanks, and cannot be used to reconstruct tool use, but they can be extremely abundant on used tools. However, they also disappear again after all the organics are consumed.

4.6.2 Mistaken identity

In the period between the appearance of the first hyphae and total breakdown of the residues, residues are still recognizable, but often with difficulty. Hyphae use enzymes to break down their resources. As a result, residues are damaged and can lose their unique characteristics.

Starch grains become opaque and dull after contact with hyphae and they lose their diagnostic extinction cross (Figures B.121 and B.125). During decomposition they are slowly incorporated into the hyphae and also lose their characteristic shape. This makes them unidentifiable and sometimes even unrecognisable.

As plant tissue is incorporated by hyphae (Figures B.126 and B.127), it loses rigid shapes and cell structure, leaving a mass that resembles muscle tissue. Figure B.128 presents a plant fibre, which resembles muscle tissue. The affected plant tissue can be birefringent. However, recent work demonstrates that muscle tissue more commonly has this attribute even though this was once thought not to be the case (Wadley, Lombard and Williamson 2004, Lombard 2005, Lombard and Wadley 2007).

Degraded plant tissue can resemble an ash or starch deposit (Figure B.129). Cell structure is lost and only many small pieces remain, sometimes consisting of small starch grains. When no starch grains are present and the particles are more cohesive, this deposit can also resemble bone.

Hyphae can resemble muscle tissue. Hyphae and muscle tissue have no clear cell structures, the material can be birefringent, but not always, and the tissue has a gelatinous constituency and can have holes in its structure. Compare the muscle tissue in Figure A.52 with hyphae in Figures B.130 and B.131. In addition, any residue that is being decomposed by hyphae is at risk for being mistaken for muscle tissue.

The starch samples often did not preserve any starch grains, but did contain pieces of epidermal plant tissue. In these experiments I knew that the plant involved was potato and that the epidermal tissue was potato skin. When bone degrades, fat becomes relatively dominant in the remaining residue. On archaeological material this bias in preservation can lead to the misidentification of processed materials.

4.6.3 Time

My results demonstrate that in order for residues to survive, the environment must be stable and favourable from the moment of deposition. Given enough time, probably within two years, there would have been no residues left on the experiments outside the cave at Sterkfontein and Sibudu and at the open-air setting at Willemina Polder and Zelhem. When residue decay is allowed to set in, it continues until all organic remains are finished. Equilibrium, as discussed in Chapter Three, cannot be reached because carbon dioxide gas is lost in the process. Residues either preserve, as inside the cave at Sibudu, or they do not. Barton (2007) also predicts a rapid initial decay of residues. From these experiments it is clear that, if a situation is not stable it takes only a year to remove most residues. Thinking in archaeological terms, this is not long.

4.6.4 Residue types

As demonstrated, some residue types are more prone to deterioration than others. Starch is extremely vulnerable. The grains are nature's energy storage cells and they are easy and rewarding for micro-organisms to break down.

Muscle tissue also preserves badly. Animal cells do not have cell walls and they are easy to break down. Although blood was not tested as thoroughly in this project as some of the other residue types, it also appears to be a fragile residue. Not one of the experiments preserved blood; not even in a sheltered micro-environment.

Bone consists of more mineral components and is much harder to decompose than other animal components. Because of the minerals in bone, the energetic reward values are low for micro-organisms compared to those for starch or muscle tissue. Fat contains a lot of energy, but because it is molecularly complex, it is also difficult to break down. Fat is often absorbed in the stone material or in the sediment and lost for analysis.

Plant tissue preserves better than any of the other tested residues. The cell walls of plant tissue do not readily decompose and plant tissue contains sugars that are more complicated and harder to breakdown than those of starch. Compared to bone-working, wood-working also leaves more residues on a tool surface, increasing the residues' survival chances.

These results have implications for starch analysis in particular. Starch grains are used to identify ancient food plants; every species has a particular shape and size that can be classified. It appears, however, that starch grains do not preserve well. Special circumstances, or a sheltered micro-environment, are required to ensure preservation. Starch grains have been extracted from grindstones and from sediment samples. Grindstones might provide a sheltered micro-environment which allows preservation. However, in the sediment, starch grains are extremely vulnerable and the validity of identifying starch grains in ancient contexts needs to be addressed.

4.6.5 Sediment types

The results presented above demonstrate that microbial activity is the key factor to residue decomposition. Particular sediment types, such as clay or ash, are better for preservation, but when microbial activity is high, a major part of residues are lost. Microbes need water, oxygen and a moderate pH to survive. Moist and permeable sediments are good for microbes. In addition, nutrient-rich sediments also have higher bioactivity than nutrient-depleted sediments. In some sediments, chemical weathering plays an important role, for example, acidic sediments do not preserve bone.

The Zelhem and Willemina Polder experiments demonstrate that in sediments with high microbial activity only few residues preserve. The clay at Willemina Polder could have protected starch grains (see Chapter Three), but in these experiments only few grains survived. It is unfortunate that the experiments in the clay sediment were not completely recovered because I cannot compare the results from these two sediment types. For now the prediction still stands that slightly alkaline clay preserves organic residues slightly better than acidic sand, but that both instigate a rapid decay process.

4.6.6 Environments

Environments play a big role in residue preservation. The results of my experiments show that open air sites preserve residues considerably less well than cave and shelter sites. Cave sites are sheltered from rain and remain dry, which leads to less bioactivity than in the open. The cave sediments at Sterkfontein are more alkaline than the sediments outside, also inhibiting decomposition. Although cave sediments contain organics that micro-organisms need, cave sediments are often not as rich as the humus layer outside the caves. These conditions lead to a slower decomposition rate inside the cave.

The experiments in the Dutch sediments did not preserve residues as well as the experiments outside the caves at Sterkfontein and Sibudu. Precipitation and bioactivity are higher and the sediments contain more sediment organics at Zelhem and Willemina Polder. Micro-organisms thrive under these favourable conditions and cause diagenesis within the sediment.

In a cave or shelter site, depending on the prevailing humidity, an exposed sample may survive better than a buried one. If the air is dry, it desiccates residues and this creates unfavourable conditions for the growth of microbes. If the air is humid, exposed samples will deteriorate quickly. It is likely that seasonality plays a role in this process. I think that the exposed starch samples inside Sibudu Cave preserved so well because they rapidly dried out during the dry season. If they had been exposed in the wet season, it is probable that they would have decayed more because they would have remained moist for longer.

Micro-environments are important for residue preservation. The sandpaper slide experiments at Zelhem demonstrate that protected residues under tools have better survival chances. In addition, some Sterkfontein tools deposited on top of the sediment outside the cave showed a differential preservation. The tool side that faced down in the sediment preserved residues better than the side that faced up towards the surface. This implies that rain has a deleterious influence on residue preservation, but that the tool itself protects the residues from decay. However, it is unclear why the exposed tools outside the Sterkfontein Cave score higher than the buried tools. If the sediment protects the tools from the elements, one would expect the buried samples to

preserve better. Perhaps microbial activity is low in the topsoil, on which the exposed tools were deposited.

4.6.7 Raw material types

The sandpaper slide experiments from Zelhem demonstrate that residues preserve badly on glass. There is no adhesion between the residue and glass surface and the residues rapidly wash off or decompose off the tools (Figure B.132). Consequently, I predict that on similar glass-like materials, such as quartz and obsidian, residues only preserve in cracks and not on the surface.

The sandpaper experiments also show that preservation on fine-and medium-grained paper is better than on large-grained sandpaper. There is a difference in grain size between the papers, but the major different lies in the quantity and spread of the grains. On the fine-grained paper, many small grains are present close to each other (Figure B.133). On the coarse paper, fewer, larger grains are located some distance from each other (Figure B.135). The medium-grained paper's results fall in between the other two papers (Figure B.134). Due to the high quantity of its grains, the total surface on the fine-grained paper is much larger than on the coarse-grained paper and this is key to residue preservation. A large, irregular surface is best for residue preservation. Future research should investigate the influence of a porous stone surface on residue preservation. Perhaps residues attach better to a porous surface, while others are soaked up in the stone and become unidentifiable.

4.6.8 Mechanical weathering

Future research should investigate whether mechanical weathering influences residue preservation. This point is closely related to reaching equilibrium as discussed in Chapter Three. I expect stable environments to preserve residues better than unstable environments. The samples inside the cave at Sterkfontein and Sibudu have better preservation than the samples deposited outside the shelters, in unstable environments.

4.7 CONCLUSION

In conclusion, residue analysis is a challenging field. Successful identifications can contribute to the reconstruction of processed materials, however, this study raises several points that analysts need to be aware of before commencing their analyses of archaeological materials. First, contaminants, particularly starch, are problematic and they need to be researched properly by examining sediment samples and by recording the distribution and association of micro-remains. If residues are well-preserved at a site they do so in large quantities and this needs to be expressed in the results. Secondly, there is a discrepancy in residue preservation. Some residues, like starch, decompose faster than others, like bone. Lastly, glass-like, light-coloured and reflective rock types can lead to a preservation and identification bias.

The contextual approach to residue analysis provides secure guidelines, which allow the identification of contaminants, use-related residues and the reconstruction of processed materials.

Site selection is important for a residue project. Most unstable open-air sites are not suitable for residue analysis. Residues need to be protected in order to survive. Micro-environments can help preservation, but they cannot compensate for a generally harsh environment.

Even if one selects a sheltered cave site for analysis, other circumstances still need to be optimal to preserve residues. The sediment should not only have little bio-activity now, but also in the past. Understanding the deposition process can lead to predictions regarding the presence or absence of residues on archaeological tools. For example, it is useful to be able reconstruct whether tools were exposed for a while or buried quickly. In the following chapters I demonstrate that a systematic and meticulous study of past as well as present decay can save time and resources by eliminating unpromising sites and their material before any residue analysis is done.

The results from the experiments have consequences for the predictions about residue preservation on the archaeological samples from Sibudu and Sterkfontein. The air at Sibudu is presently humid because of sea-aerosols and some moisture is available for the micro-organisms that live in the top layer of the sediment. Fungi and bacteria are abundant in this part of the sediment and they are probably not, or at least not active

in the rest of the sediment. This leads to a higher decomposition rate at the top of the Sibudu deposit.

For the archaeological sample this implies that there is not good residue preservation in occupation surfaces that were exposed for long periods. The Sibudu sediments are deposited with long hiatuses between the age sets (see Chapter Three). This means that residue preservation is probably less at the top layer of these age sets, because they were exposed to the humid air for long periods. I predict that the Grey Rocky, Brown Speckled, Mottled Deposit and Coffee layers preserve fewer residues than the other layers. The pre-60 ka period was more humid than the 60 and 50 ka occupations; I therefore also expect fewer residues in these layers. The 60 ka and 50 ka sediments accumulated rapidly (the sequences are very deep although each accumulated in less than 1000 years) and the sediments indicate that the environment was probably less humid than nowadays and also less humid than the pre-60 ka period. More residues are expected to survive in the 60 ka and 50 ka layers than in the pre-60 ka layers; there maybe even better preservation than the present day. The absence of gypsum in the pre-60 ka levels needs addressing to allow confident predictions.

On the sample deposited outside the cave at Sterkfontein only few residues preserved after one year. The microbials in the sediment successfully attacked the residues. Inside the cave, where it is dry and where microbial activity is low, residues preserved well. This can imply that residues are also present on the archaeological sample if it was originally deposited inside the cave.

During the M5 infill the climate was warmer and slightly drier than present day. The clay and silt in the sediment matrix indicates that a stable palaeosoil was present outside the cave. The tools were deposited on this stable soil and only washed into the cave during catastrophic events towards the end of the interglacial. Therefore the tools may have been exposed outside the cave for a long period during the interglacial. Even if the environment was warmer and slightly drier than present day, exposed residues still deteriorated rapidly. There are probably no ancient organic residues on the archaeological tools from Sterkfontein. In the following chapter I test these predictions.

Future research should focus on preservation of residues in different sediments and soils and refine the preservation predictions from paragraph 4.7. Investigation into living floors or stable palaeosurfaces will probably demonstrate that well-developed

palaeosoils, which are predominantly formed in humid and warm (pluvial/interglacial) periods do not preserve micro-remains (Strahler and Strahler 1997, Rapp and Hill 1998). Poorly developed palaeosoils and colluvial surfaces, which occur in drier and cooler than present conditions (interpluvial/glacial), will probably preserve residues better (Strahler and Strahler 1997, Rapp and Hill 1998).

5 RESULTS FROM THE STERKFONTEIN ARCHAEOLOGICAL SAMPLE

In this chapter I describe and discuss the results from the residue analysis of the archaeological sample from Sterkfontein. The results are presented in tables and Appendix C provides distribution maps of the residues on the tools and micrographs of the residues. The main objective of analysis is to investigate the possible correlation between the preservation of experimental and archaeological residues at Sterkfontein. Because Dr Tom Loy analysed the sample in the past, it also gave me an opportunity to revisit part of the collection that he worked with. Several predictions for residue preservation were formulated in the previous chapter, and this chapter attempts to test these predictions against the archaeologically recovered material.

The variables that are of interest are time, climate, sediment and environment and they differ greatly between the Sterkfontein and Sibudu archaeological sites, which is why these sites were selected. The material from Sterkfontein is much older than Sibudu and the material at Sterkfontein was deposited around the cave entrance and was therefore exposed rather than protected in a cave, as was the case at Sibudu. The sediment in which the tools were deposited was humic at Sterkfontein and of non-anthropogenic origin, but at Sibudu the sediment is dry and consists of anthropogenically formed ash and sediment.

The experimental lithic blanks and the used, replicated tools deposited at Sterkfontein are discussed in detail in Chapter Four. These tools acquired some organic contaminants from the sediment matrix outside Sterkfontein Cave. These contaminants are mainly plant fibres, insect remains and pollen that are naturally present in the Sterkfontein sediment samples. The preservation of the experimental residues outside the cave was poor after one year and I therefore predicted that the archaeological tools would not retain ancient residues.

Professor Ron Clarke and Dr Kathleen Kuman excavated the archaeological tools, which were analysed for residues by Dr Tom Loy between 1998 and 2001, from Sterkfontein in the 1990s. Loy found organic residues on several flakes and cores and inferred that the lithics had been used to work bone, muscle tissue, connective fibres

and plant materials (Loy 1998a). He conducted DNA analysis on blood found on some of his analysed tools and he successfully amplified DNA in ten cases. The blood samples came from formal tools (retouched flakes and cores/bifaces). He concluded that in four instances hominid blood was present on these tools (STK3347, STK2503, STK6122 and STK10363), but in the six other cases he may not have completed analysis (STK2371 two times, STK3854, STK3932, STK3977 and STK8911) (Loy pers. comm. 2002). Dr Bonny Williamson was also able to amplify DNA found in a blood residue on a Sterkfontein tool (STK10091). She cautiously concluded that hominid blood was present. Williamson and Loy's DNA sequences of the hominid blood were exact matches, even though the amplification was conducted at two different laboratories (Williamson 2000b). Unfortunately, Loy's results are not published and there are no images or data available of the residues or DNA that he identified. His study of blood residues is sometimes considered to be controversial (Renfrew and Bahn 1996).

Although Loy has not published his DNA results, he presented some other results in 1998 at the Dual Congress conference at Sun City, South Africa. He and also prepared several publications and reports, which I was able to read. In 2002 Loy gave me a part of his analysis documentation. When needed, Kuman gave me additional information on his study. I used these documents and personal communications to reconstruct Loy's analysis.

5.1 RESULTS

I analysed 27 flakes, two retouched flakes, two core tools and one core. Nine of these pieces were also analysed by Dr Tom Loy. The results are summarized in Table 5.1. If precise locations of micro-remains are not mentioned in the descriptions below, then there were no patterns of any interest.

STK403 (Figure C.2)

The quartz flake did not contain many residues. There are some blue fibres and two black deposits present on the dorsal side. The flake also has several red/orange sediment spots and one yellow-orange deposit, covering the distal end of the dorsal

side. This yellow-orange deposit is very similar to the nail varnish found on STK 2504.

STK2053 (Figures C.3 and C.6)

Many red breccia and black deposits occur on this small chert biface. In addition blue plant fibres and a mint-green, fat-like deposit are also present. Fat and fatty bone are found all over the tool, most in correlation with sediment patches (Figures C4, C.7 and C.8). The expected working edges, the sides and tip, contain few micro-remains. The ventral side contains many ink patches (Figure C.5). In addition, this tool was cast and the silicon rubber that was used is greasy and fatty. It is most likely that the rubber left micro-remains on the tool surface in the form grease/fat deposits.

One fat residue near the tip of the biface is covered with a light sediment layer. This fat deposit could be an ancient use-related residue. However, it is the only unambiguous residue and there is no correlation with other micro-remains that could reveal what this biface was used for. From the experimental tools in Chapter 4 I learned that residues always occur in patterns and in correlation with other residues. The maps in Figures C.3 and C.6 illustrate the random distribution of the residues. Therefore, I consider the fat deposit on STK2053 not use-related.

STK2504 (Figure C.9)

This quartz cortical flake still contains much sediment. The contaminants present include black spots and large areas with ink deposits. One orange-yellow deposit, illustrated in Figure C.10, includes air bubbles. This is a resinous deposit, but probably a contaminant nail varnish from labelling. This deposit is located on the obtuse side of the tool, close to the label. I found no organics such as plant fibres in connection with it.

Loy analysed this tool and found red blood cells, nucleated cells and cellulose. He concluded that the tool was used for cutting muscle tissue and plants. He also noted that the resinous patch on the lateral ventral edge is probably nail varnish. Figure C.9 shows that no animal residues or cellulose are present. The flake is covered in ink deposits, clearly illustrating the compromised curation.

STK2630 (Figure C.11)

This small incomplete quartzide flake contains only sediment and black spots. The distribution map shows the random distribution of sediment and black stains on the ventral side of this flake.

STK2739 (Figure C.12)

This larger quartzite flake contains some organic material, which is mainly distributed in and around sediment patches. Plant fibres are present over the entire surface of the tool. Bone and fat are present in association with sediment. Rootlets and one instance of insect remains occur, but no spores, pollen or hyphae are present. The orange sediment patches that are associated with the organic contaminants are spread out over the entire surface and not focussed around the edges.

STK2808 (Figure C.13)

Some plant fibres are present in the sediment matrix. Several fatty bone-like flakes are visible on the quartz flake's dorsal ridge on top of sediment patches. These are probably skin fragments that come from recent finger rubbing. Black deposits are visible all over the flake surface. Figure C.14 and C.15 show a plant fibre on top of a sediment patch and a typical black deposit and sediment spot respectively.

STK2982 (Figure C.16)

This quartz incomplete flake contains some obvious contaminants, such as bright blue fibres and ink smears. The remains are randomly distributed and Figures C.17 and C.18 show contaminant fibres on top of sediment patches. Some hyphae are present, but no spores.

STK3049 (Figure C.19)

This chert flake contains no organic material. Some red sediment patches and black spots are present and also a blue granular deposit (Figure C.20). On the distal end of the dorsal side is a yellow-orange nail varnish deposit present as in STK403 and STK2504.

STK3051 (Figure C.21)

On the quartzite flake I found evenly distributed sediment patches and black stains. One bone-like residue is present, but it is in association with a sediment patch (Figure C.22). Loy noted that this flake contains hairs and fatty bone remains. I did not find any hairs and the bone residues are incorporated in the sediment.

STK3067 (Figure C.23)

This quartz incompelete flake contains sediment and black patches that are often mixed together (Figure C.24). A number of plant fibres are present on the dorsal side, some are found on top of a sediment patch and similar ones are found away from the edge of the flake.

STK3109 (Figure C.25)

The small quartz flake has some areas where the surface is red/pink in colour. Although this mimics a deposit it is part of the stone surface. One bright white contaminant plant fibre is present on the dorsal side.

STK3238 (Figures C.26 and C.27)

This large retouched chert flake contains many sediment and black patches and contaminant plant fibres. There are several bone-like deposits, but they are always on top of, or associated with sediment of black stains and they are probably part of the sediment matrix (Figures C.28 and C.29). It is clear that the spread of micro-remains on the surface is random.

STK3312 (Figure C.30)

The distal end of this quartz flake is broken. It contains many blue fibres and one mint-green deposit; all of these are obvious contaminants. A few white fibres are present on the dorsal and ventral sides, together with red sediment deposits, some of which are deep red in colour. Loy identified red blood cells in some of the tool's cracks, but I could not find them. One putative blood deposit that he described looks more like a typical sediment accumulation than a blood deposit. I did not find any bone residues.

STK3363 (Figure C.31)

This quartz flake contains no micro-remains, but several black deposits and sediment areas on the ventral side are present. The black deposits are either manganese (Figure C.32) or ink (Figure C.33).

STK3411 (Figures C.34 and C.35)

The ventral side of this quartzite flake is not useful for residue analysis because it is covered in writing and is therefore too disturbed. The dorsal side shows many patches with plant fibres, but they are always associated with sediment (Figures C.36 and C.37).

STK3977 (Figure C.38)

This quartz core contains many black and red sediment deposits. Several bright fat-like deposits are present (Figure C.42). There are fatty bone-like deposits present (Figure C.39) and these are explained by handling and casting, which must have left bone-like skin flakes and fatty deposits on the surface. The yellow areas that occur (Figures C.40, C.41 and C.43) are not deposits, but inclusions. Figures C.40 and C.41, taken at the same spot in normal and cross-polarised light respectively illustrate that the yellow areas are part of the stone.

STK4883 (Figure C.44)

This very small quartz flake contains some white and blue fibres. The white fibres are similar and always associated with sediment (Figure C.45). Some orange sediment deposits occur on the ridges.

STK5872 (Figures C.46 and C.49)

All the micro-remains that I identified on this large quartz flake are also present on top of sediment patches and there is no pattern in the distribution of the micro-remains. Because they occur on top of the sediment, they must be modern contaminants. These residues include white plant fibres (Figure C.50), fatty bone (Figure C.52), fat (C.47 and C.48) and fat-like deposits (Figure C.51 and C.54) and black accumulations. There is a wide variety of contaminants present on this tool; some like the bright fat-like deposit in Figures C.51, C.53 and C.54 are obvious and present all over the surface. Loy commented that this tool had a “collagen sheet” on its thin edge and that

it also contained a fat deposit. He concluded that the flake was probably used as a spokeshave or scraper on fresh, dry bone. I did not find an extensive bone deposit or any other unambiguous residue. Kathleen Kuman (pers. comm. 2007) examined the tool and concluded that there is no use-wear. Spokeshaves and scrapers display much use-wear and it is therefore unlikely that that the tool identification by Loy is correct. Some of the fatty bone I found was positioned on top of sediment patches or ink (Figure C.52) and the distribution pattern of bone is random. In addition, the tool is clearly contaminated with modern remains and this compromises all micro-remains. Therefore, all residues including fatty bone are dismissed.

STK5885 (Figure C.55)

This small quartz flake has a partly broken distal end (see the dotted side in Figure C.55). This flake contains many fatty bone flakes. However, they are located near the broken edge and in most cases they are associated with sediment (Figure C.56). From the experimental sample I learned that bone and connective fibre residues are often confined to the edge. Because the bone was found away from the possible working edge and in association with sediment, these micro-remains must be contaminants. I also found some blue contaminant fibres. Loy supplied two short records for this flake and it is not clear what exactly he was referring to. In one record he states that this tool was used for cutting plant material and in the other it says “possible contaminants”. Since I did not find any plant residues I assume that he also concluded that this sample only contained (sediment) contaminants.

STK6048 (Figures C.57 and C.59)

This quartz sample contains many black mineral inclusions and black deposits that are best observed along the edge and on the dorsal side. The tool contains some fatty bone remains, often associated with sediment (Figure C.58). The plant fibres that are present (Figure C.60) are also associated with sediment. In addition, several ink patches occur (Figure C.61) and I deduce that no use-related residues occur on STK6048.

STK6049 (Figures C.62 and C.64)

This quartz flake is similar to STK6048. It contains sediment and black spots on which some plant fibres are present. It also shows the same black deposits in cracks as

STK6048. These deposits are located directly next to the label and are probably ink. Because this quartz tool is so light, it clearly shows the almost liquid nature of breccia (Figure C.63). Without careful examination of sediment samples and all deposits on many Sterkfontein artefacts, this kind of sediment can be confused with resin or another liquid residue. There are some fatty bone and fat deposits, but the distribution is random and they are often associated with sediment (Figure C.65).

STK6085 (Figures C.66 and C.74)

This large chert flake is covered in red sediment. The tool contains many bone-like flakes and bright, white plant fibres. Almost all of these organic remains are associated with sediment and are found in the interior of the tool, away from the possible working edge. The few that are not are extremely similar to the ones that are definite contaminants. Loy identified this flake as a spokeshave that was used in bone working. He notes bone deposits and connective fibres. The possible bone-like flakes and connective fibres I found are most likely skin flakes from handling (Figures C.67 and C.68). Dry skin cells contain collagen and probably have similar qualities as bone flakes. Kathleen Kuman (pers. com. 2007) could not identify any use-wear, commonly associated with spokeshaves. In addition, the bony, skin flakes I found are randomly dispersed over the surface or associated with sediment (Figures C.66 and C.74). Perhaps Loy misidentified the bright white plant fibres as connective fibres (Figure C.69). Connective fibres can be birefringent, but not to this extent. Bone also does not show cell walls, as these fibres do. In addition, the fibres all seem to float over the surface, indicating a recent deposition. STK 6085 also contains several fat deposits (Figures C.71 and C.73), which are associated with sediment and are randomly distributed. Figures C.70 and C.72 illustrate the extensive presence of ink and contamination on this tool.

STK6121 (Figure C.75)

This quartz flake contains sediment and black deposits. On the dorsal side are some hyphae remains present. Loy also analysed this tool and also found no residues.

STK6129 (Figure C.76 and C.80)

This quartz flake contains many black and iron stains. Some starch grains are present, but they are clearly contaminants because they are present on top of the nail varnish

of the label (Figures C.77, C.79 and C.81). A bright plant fibre and a fat deposit (Figure C.78) are also present, but they have an arbitrary distribution. Loy analysed this tool, but I could not obtain the records of his analysis.

STK6230 (Figures C.82 and C.85)

The dorsal side of this large cortical quartz flake was covered in black stains and sediment deposits. Both ventral and dorsal sides contain bright plant fibres and some bone-like skin flakes and all of these contaminants are found in association with sediment or black patches. Other contaminants are nail varnish (Figure C.83) and a blue plant fibre (Figure C.84). Loy analysed this flake, but the records of this tool are not available.

STK7318 (Figures C.86 and C.87)

This chert flake contains some thick and large sediment patches. Plant fibres are spread equally and randomly over the dorsal surface. Some of these fibres are present on top of sediment (Figures C.88 and C.89) and they are clear contaminants.

STK7975 (Figures C.90 and C.91)

This chert flake contains many similar bright white contaminant fibres. They are present on the dorsal and ventral sides and over the entire surface of the tool. Some of these fibres were found on top of the nail varnished label, on top of sediment patches and near unlikely use edges, such as the thick and obtuse proximal part. The bone remains on the dorsal side are on top of sediment and ash deposits and therefore clear contaminants. The bone on the ventral side is located in the interior of the flake, without a clear use pattern. The flake is heavily stained with black deposits, which are visible with the naked eye.

STK8075 (Figure C.92)

This small quartz flake shows black residues and sediment staining and some ash patches. One fat-like residue occurs on top of a sediment patch on the dorsal side and is dismissed as a contaminant. The bone fragment on the nail varnish on the ventral side is also a contaminant. The fat on the ventral side is an isolated single residue that, without associated residues and a use-related distribution pattern, must be discarded as incidental.

STK8849 (Figure C.94)

Because the ventral side was covered in writing (Figure C.93) and clearly too contaminated, I only analysed the dorsal side of this quartzite flake. Besides sediment deposits and black staining, I found some white ashy deposits, which were incorporated within the sediment patches. These deposits are thin and cover most of the tool.

STK8855 (Figure C.95)

In addition to the usual black stains and thin sediment patches I found some brightly coloured purple plant fibres, which emphasise the contaminated nature of this flake. The bone deposit on the dorsal side is an isolated residue and was therefore disregarded.

STK8903 (Figure C.96)

This small quartz flake with retouch on its distal end contains one sediment patch, but no other contaminants or residues.

STK9521 (Figures C.97 and C.101)

This large chert flake is covered in uniform white plant fibres, which are found in small clusters or as single fibres. Many times they are associated with sediment patches (Figures C.98, C.99 and C.100). These fibres are clearly modern contaminants. The few fat residues present have a random distribution and are often associated with sediment (Figure C.102).

STK10363 (Figure C.103)

Due to its size, this large quartzite cleaver did not fit well under the microscope and I was only able to analyse the distal end. This end is the most likely working edge. There is a random spread of blue, white and brown fibres over the analysed surface, confirming that this precious piece was wrapped in textile for protection (personal observation, 2005). The tool contains many areas with fat or grease, bone-like remains and black stains. Loy reported that he found blood residues along the cutting edge. There are red deposits around the tool edge, but these are the same sediment deposits that are found all over the tool (Figure C.103). In some cases I found fat on top of the sediment spots (Figures C.105, C.106, C.107 and C.108). This tool was cast

and the silicon rubber that was used is greasy and fatty. It is most likely that the rubber left micro-remains on the tool surface in the form of grease or fat deposits.

Table 5.1: Overview of the Sterkfontein archaeological samples and analyses; Co= contaminants only.

Tool number	Tool type	Deposit	Raw material	Cast	Loy's residues and interpretation	Langejans's residues
STK403	Flake	M5E-A	Quartz	-	-	Co
STK2053	Biface	M5E-A	Chert	Yes	-	Co
STK2504	Cortical flake	M5E-A	Quartz	-	Red blood cells and cellulose.	Co
STK2630	Incomplete flake	M5E-A	Quartzite	-	-	Co
STK2739	Flake	M5E-A	Quartzite	-	-	Co
STK2808	Incomplete flake	M5E-A	Quartz	-	-	Co
STK2982	Incomplete flake	M5E-A	Quartz	-	-	Co
STK3049	Flake	M5E-A	Chert	-	-	Co
STK3051	Flake	M5E-A	Quartzite	-	Bone and hair	Co
STK3067	Incomplete flake	M5E-A	Quartz	-	-	Co
STK3109	Flake	M5E-A	Quartz	-	-	Co
STK3238	Retouched flake	M5E-A	Chert	-	-	Co
STK3312	Flake	M5E-A	Quartz	-	Blood with red blood cells visible and bone.	Co
STK3363	Flake	M5E-A	Quartz	-	-	Co
STK3411	Flake	M5E-A	Quartzite	-	-	Co
STK3977	Core	M5E-A	Quartz	Yes	Blood. Successful ancient DNA sequencing, unknown donor.	Co
STK4883	Flake	M5E-A	Quartz	-	-	Co
STK5872	Flake	M5E-A	Quartz	-	Bone and fat indicate use as a spokeshave or scraper on dry fresh bone.	Co
STK5885	Flake	M5E-A	Quartz	-	Contaminants.	Co
STK6048	Flake	M5E-A	Quartz	-	-	Co
STK6049	Flake	M5E-A	Quartz	-	-	Co
STK6085	Flake	M5E-A	Chert	-	Bone and connective fibres indicate bone working.	Co
STK6121	Flake	M5E-A	Quartz	-	No residues	Co
STK6129	Flake	M5E-A	Quartz	-	?	Co
STK6230	Flake	M5E-A	Chert	-	?	Co
STK7318	Flake	M5E-A	Chert	-	-	Co
STK7975	Flake	M5E-A	Chert	-	-	Co
STK8075	Flake	M5E-A	Quartz	-	-	Co
STK8849	Flake	M5E-A	Quartzite	-	-	Co
STK8855	Flake	M5E-A	Quartz	-	-	Co
STK8903	Retouched flake	M5E-A	Quartz	?	-	Co
STK9521	Flake	M5E-A	Chert	-	-	Co
STK10363	Cleaver	M5E-A	Quartzite	Yes	Blood. Successful ancient DNA sequencing, hominid donor.	Co

5.2 DISCUSSION

I did not find convincing evidence for ancient residues on the Sterkfontein tools. This result differs from earlier work by Loy. My approach was different from that of Loy: my analysis took cognizance of residues that are naturally within sediment samples and my comparative collection included experimental tools some of which were without residues (labelled blanks) as a control sample. I also looked at residue distributions and their associations on each tool. My analysis was strict and all ambiguous or single and isolated residues were discarded. Ambiguous residues include those types of residues that are present within sediment samples and on blank tools. Other residues such as those on top of sediment patches or on nail varnish used for labels are also considered to be contaminants.

The sediment and breccia samples demonstrate that plant fibres and tissue, hyphae, pollen, charcoal, rootlets and insect remains are part of the sediment matrix and are likely to contaminate artefacts. Compare Figures A.99, C.14, C.18, C.37, C.45, C.50, C.88, C.89, C.98, C.99 and C.100. The plant fibre from the sediment matrix (Figure A.99) is similar to the fibres on the ancient tools and the fibres on the tools are also similar to each other. Fat-like deposits on the ancient Sterkfontein tools as in Figures C.7, C.8, C.47, C.48, C.73, C.78, C.102, C.104, C.105 and C.108 are also present on blanks (Figure B.21) and on breccia (Figures A.105-A.107). Fatty bone-like residues are common on the ancient Sterkfontein tools (Figures C.4, C.28, C.29, C.56, C.65, C.67, C.68, C.78, C.106, C.107). Fatty bone-like remains are not abundant in the sediment matrix or breccia, but present nonetheless (Figure A.101). In general the fatty bone-like remains are strikingly similar on all ancient tools.

In Chapter Four I demonstrated that connective fibres, fatty bone and wood residues are often confined to the used edges of experimental tools. However, in this archaeological sample I found bone-like flakes or skin flakes, fat and woody residues over the entire tool surface, which is a typical distribution for contaminants (as is an isolated, single residue). These contaminants are the result of handling and storage. The woody tissue and plant fibres originate from the sediment, paper storage bags or protective cloths. Fat and fatty bone-like flakes derive from handling and casting. Finger grease and skin flakes consist of the same building blocks, lipids and connective tissue, and they have the same properties as archaeological fat and bone.

In general, this study did not look at single possible residues and its properties, but complementary to Loy's study, it also looked at residues within the tool and sediment contexts.

It is problematic to apply residue analysis to the archaeologically recovered Sterkfontein tools. Their curation is, as explained in Chapter Two, complex. They were handled extensively before Loy's and my analyses, leaving bone-like skin flakes, finger grease, plant fibres, starch grains and other contaminants on the tools. Residue analysis should never be applied to tools with a post-excavational history such as that of the Sterkfontein sample.

Loy claimed to have found several blood residues (Loy 1998; Loy pers. comm. 2002). It is possible that he removed these to conduct Hemastix or Chemstrip tests as he regularly did so other studies (*e.g.* Loy and Dixon 1998). However, there is no reference to this practice in his notes. Loy certainly removed a possible blood residue from sample STK3977 for DNA analysis. Such removals could explain why I did not find these residues. However, even if Loy extracted the residues and positively identified them, caution is required. Hemastix and Chemstrip tests are known to give false positive results, particularly at archaeological sites like Sterkfontein, which have abundant manganese-oxide deposits (Custer, Igenfritz and Doms 1988, Manning 1994, Downs and Lowenstein 1995). The use of Hemastix and Chemstrip tests and the crystallisation of haemoglobin, which Loy (1983) also might have used, should be "regarded with healthy scepticism" (Smith and Wilson 2001, 315) (for an overview of the discussion regarding the analysis of blood residues, see Smith and Wilson 2001). Another reason for being cautious about the putative presence of blood on the tools that Loy examined is that animal remains, such as blood, seldom occur as isolated residues. Results from the experimental sample show that animal remains usually occur within suites of residues that include at least some bone, muscle tissue, connective fibres or fat. Isolated animal residues (and, indeed, isolated plant residues) should be treated with caution and should be considered unreliable (for a complete overview of this argument, see Chapter Two and Lombard and Wadley 2007, Wadley and Lombard 2007).

The Sterkfontein tools are strikingly similar; all contained orange to deep-red sediment deposits, black stains, and red or black cast-resin spots, some white ash, white and brightly coloured plant fibres, some pollen and most of them also contained

bone-like and fat deposits. These remains are spread randomly over the full surface of the samples and show no patterning. All samples, regardless of their tool type, size and raw material have the same contamination pattern. If these micro-remains were ancient residues, one would expect to see use patterns within, for example, the retouched flake group.

Early hominids deposited the Sterkfontein tools around the cave entrances. Over time these tools washed into the caves. It is unclear how long this erosion process took, but the experimental sample, also deposited around a cave entrance, had not washed into the cave after one year. Most of the experimental residues, however, were already lost by then. In addition, the stable paleosol and perhaps the manganese stains on the tools could indicate that the tools were exposed outside the cave for a considerable time. Once the archaeological tools washed into the cave and onto the talus cone, the sediments calcified and formed a solid, hard breccia. Over time, a part of the breccia decalcified again and became soft once more (Kuman and Clarke 2000). Jones (1998) concludes that the circumstances inside Sterkfontein Caves were favourable for residue preservation due to the clay fraction and calcide ions in the sediment, which binds and seals the organic residues and in doing so protects them. My result from the experimental sample of tools that was buried outside the cave entrance could confirm her analysis. The residues on the buried sample of replicated tools are better preserved than the ones on the tools that were left exposed on the surface. However, the buried sample of residues is still extremely deteriorated. Jones's analysis (Jones 1998) is based on the sheltered situation inside the cave, but because the tools were initially deposited outside, it is this outside environment that dictates the potential for residue preservation. The climate at the time of deposition was slightly warmer and perhaps more wet (due to more winterrainfall) than present day (Kuman and Clarke 2000, Avery 2001), and even if the archaeological tools were only exposed around the cave entrance for five years, that was probably enough time for all of the organic residues to be removed by the elements. It is important to remember that Sterkfontein is not a cave site in primary context.

My observations leave a question mark over Williamson (Williamson 2000b) and Loy's (Loy pers. comm. 2002) discovery of hominid DNA on four Sterkfontein samples. The hominid DNA from these tools is identical (Williamson 2000b).

Williamson removed a blood sample from the Early Acheulean cleaver (STK10091) by dissolving it in pure water. She used a pipette to remove the solution from the surface. Williamson mentions that she used the same technique as Loy did, although his work on samples STK3347, STK2503, STK6122, STK10091 and STK10363 is not published (but see for his methods on DNA analysis see Loy 1993). Williamson's samples were analysed at the Biochemistry Department at the University of the Witwatersrand in Johannesburg. The extraction was conducted in an isolated room, but this laboratory is not a clean or sterile laboratory. No replications were made and the samples were not sent to other laboratories for control testing. Loy conducted his analysis at the Centre for Molecular and Cellular Biology, University of Queensland in Australia.

Because part of the Sterkfontein tools are currently reanalysed and stored at the Sterkfontein laboratory on site, I was unable to examine STK10091. However, I analysed STK2053, a biface on which Loy also found blood residues, STK3977, a core from which Loy was able to sequence DNA and STK10363, a cleaver from which Loy was also able to sequence DNA (Loy pers. comm. 2002). The results from my microscopy on these tools are summarised above and, in short, I did not find any blood residues.

When analysing DNA from a tool surface Hardy, Raff and Raman (1997) recommend that the sediment surrounding the sampled tool and the sediment from other parts of the excavation also be tested for DNA. If the results from the tool and sediment are similar then there could be a bias. Cooper and Poinar (2000) published some key criteria to exclude contamination in DNA analysis (Also see Handt *et al.* 1994):

1. Physically isolated work area for ancient DNA.
2. Control amplifications.
3. Appropriate molecular behaviour.
4. Reproducibility.
5. Cloning.
6. Independent replication.
7. Biochemical preservation.
8. Quantitation.
9. Associated remains.

Hofreiter *et al.* (Hofreiter *et al.* 2001, Leonard *et al.* 2007) add that DNA analysis on soft tissue is particularly complicated and that one should be extremely cautious with human DNA. The chance for contamination with modern human DNA is immense and Hofreiter *et al.* (2001) demonstrate that extensive analysis is needed to differentiate ancient and modern human(oid) DNA, but that this is often conducted. Furthermore, they state that material older than 100,000 years old is too deteriorated for analysis (also see Ambler and Daniel 1991, Eglinton and Logan 1991, Poinar and Stankiewicz 1999). In addition, most DNA samples are extracted from inside a bone piece to avoid contaminants on the outside of the bone. The Sterkfontein DNA samples were extracted from the surface and it is likely that modern human DNA was abundant on the Sterkfontein tools, which were extensively handled for years before the analysis was performed. DNA deteriorates rapidly after an object is excavated and water cleaning also greatly decreases the DNA yield (Pruvost *et al.* 2007). There was considerable time, more than 10 years, between when the Sterkfontein artefacts were excavated and when they were sampled for DNA.

Williamson did not have the opportunity to meet most of the Cooper and Poinar (Cooper and Poinar 2000) requirements and she also did not analyse control sediment samples. Pipetting the blood solution from the tool in a "dirty" laboratory allowed for more windows of error. In addition, both Williamson and Loy's samples are too old to preserve DNA according to research on the subject. The results are also problematic because they analysed a soft tissue. Because both Williamson and Loy's samples appeared to be hominid/human, extra caution should be exercised. Human DNA is a common contaminant and although the sequence did not appear to be from a modern human (Williamson 2000b), it could very well be a damaged sequence. Although Williamson explains the identical results as confirmation of the credibility of the analyses, she is cautious about the results. The identical sequences can also be explained by presence of the same contaminant. Thus, Williamson and Loy probably analysed modern contaminant human DNA.

5.3 CONCLUSION

The Sterkfontein tools did not yield convincing evidence for the presence of ancient residues and the discrepancy between my results and Loy's results can be explained

by a difference in method. Had Loy conducted experimental work, analysed sediment samples from Sterkfontein and paid more attention to the distribution and association of the putative residues that he observed, he probably also would have discarded them as contaminants.

My negative results from the archaeologically recovered tools are to be expected because of the results from my experimental sample. The experimental tools were deposited outside the cave where microbial activity is high. Outside the cave mechanical and chemical weathering, caused by rain, wind and sediment movements, also play an important role. Thus, after one year, few experimental residues preserved. The archaeologically recovered tools would have been subjected to the same destructive processes. The curation history of the Sterkfontein archaeological sample shows that the tools were prone to contamination. As a further complication, most of the archaeological tools were made on quartz. Quartz is highly reflective and is a poor rock for residue preservation and analysis of residue distributions. Glass-like materials like quartz do not preserve residues well, other than in cracks.

From the discussion above, it might appear to the reader that residue analysis is not replicable and therefore not reliable. Analysts could be accused of reaching different conclusions about the presence or absence of residues on the same tools; however, this is not the case when analysts use the same stringent methods. Blind tests by Wadley, Lombard and Williamson (2004) and more recently by Wadley and Lombard (Wadley and Lombard 2007, Lombard and Wadley 2007) show that although residue analysts are still grappling with some identification issues, their results can be replicated and their experimental work is providing credible results.

The following chapter, which summarises the results of residue analysis at Sibudu Cave, provides greater standing for the technique of residue analysis than the Sterkfontein results.

6 RESULTS FROM THE SIBUDU ARCHAEOLOGICAL SAMPLE

6.1 INTRODUCTION

In this chapter I describe and discuss the results from the residue analysis of the archaeological samples from Sibudu Cave. The results are presented in tables, photographs and distribution maps and in Appendix D. From the archaeological sample I want to learn how well residues preserve at Sibudu and if there is a bias in residue preservation of residue types. In addition, I want to explore the possibility that there is a correlation between the preservation of experimental and archaeological residues. The variables that are of interest are time, climate, sediment and environment. The artefacts from Sibudu, with an age of about 70 and 60 ka old, are much younger than those from Sterkfontein and, unlike the secondarily deposited Sterkfontein artefacts, the material at Sibudu was deposited in primary context inside the shelter and was therefore protected rather than exposed, as was the case at Sterkfontein.

The experimental material from Sibudu shows that residues preserve well minimally over a period of one year. The ancient residues may preserve better than modern ones because the shelter would have been farther from the coast, and therefore protected from humid sea aerosols, during the last glacial. The overhang provides protection from most climatic influences and the dry, anthropogenic and ashy sediment also prevents speedy biodegradation of organic micro remains. The drier environmental and similar physical situation of the ancient tools, compared to the experimental sample, leads me to predict that residues should preserve well at Sibudu Cave and that ancient remains with depositional integrity are expected on the archaeologically recovered stone tools. This chapter attempts to test these predictions.

The tools were collected specifically for this study during the 2007 field season. Only a limited number of people excavated and handled the tools, and the tools were immediately secured in plastic ziplock bags to avoid contamination. After the field

season, the tools were stored at the University of the Witwatersrand and not handled by anyone besides myself. Wadley and her team collected 30 possible *pièces esquillées* (scaled pieces; labelled SBL1-SBL30) without handling them. Paola Villa (pers. comm. 2007) classified eight pieces as definite *pièces esquillées* and the remaining artefacts are flakes and pointed flakes. During her identification the artefacts remained in the plastic bags.

6.2 RESULTS

With regard to the results, several points need clarification. Most tools contained sediment and were brushed lightly before analysis because residues could not otherwise be seen. However, I analysed all tools before and after brushing, ensuring that no data were lost. I observed that residues are not dramatically affected by brushing, but this action sometimes leaves brush marks on the surface of the tool and these must not be confused with use marks (Figure D.142). In addition, loose and weightless remains, such as fibres and starch grains can move around during brushing, but residues, which are attached to a tool surface, such as resin and fat, are not affected.

Fat and burnt bone and bone fragments are abundant in the assemblage and they do not always appear to be use-related. The remains are strikingly similar on all the artefacts. Fat consists of bluish-white deposits or smears and is not granular at higher magnifications. Under polarised light, fat is opaque and often turns a dark blue. Under cross-polarised light it can be slightly birefringent. Burnt bone ranges in colour from yellow to dark orange and it has a number of characteristics in common with fat and unburnt bone and connective fibres. In some cases the bone deposit consists of small pieces or flakes and in other cases it appears as a viscous smear (also see Chapter Two). Some bone deposits are present on top of sediment and ash layers and I identify those as ancient, but post-depositional because many tiny bone flakes occur in the archaeological deposit and burnt bone flakes occur in the ancient hearths (Figure D.129). Other bone deposits occur under sediment and ash and I identify these as ancient use-related residues. In some cases it is unclear whether bone residues are use-related or not and in these cases I look at their association with other animal type residues, such as muscle tissue, connective fibres and fat deposits. If other animal type

remains are associated with the burnt bone and fat, and if there is a meaningful and repetitive distribution, then I conclude that the residues are use-related. If the fat and burnt bone remains are isolated and appear random, then I identify them as unintentional or post-depositional residue contaminants. The fat and burnt bone deposits, which appeared purposefully smeared or pressed onto the surface, are also identified as ancient use traces. Modern contaminants and post-depositional residues are unlikely to be forced onto a tool, but residues from tool use probably are. I cannot state categorically how fat and burnt bone could have appeared on the artefacts as non-humanly derived residues, but post-depositional factors provide the most likely explanation. These include: accidental burning in the hearth or close to the hearth. In such situations fatty bone splinters that were previously smashed open for marrow, will release fat and are likely to crumble into fragments. Burnt bone and fat are part of the sediment matrix and pieces can adhere, with sediment, to the tool. In addition, fatty residues on tool surfaces could have resulted from handling with greasy hands during ancient times. To illustrate the abundance and often random distribution of fat and burnt bone, I illustrate all ancient use residues and what I consider accidental residues or ancient contaminants in the distribution maps in Appendix D. The nature of these residues on the tools is discussed in the text.

Ancient residues are not necessarily use-related and it is important to note that unused flakes are part of a dynamic tool sequence. For example, hands of the people who lived in the shelter in prehistory were probably quite dirty and artefacts can contain unintentional residues simply from handling. In addition, unused flakes are deposited in the living area and it is likely that accidental spatters and drips fell on the tools. These residues are genuine and ancient, but unintentional and not use-related. They are part of the normal “background residues”, which are present on all tools as a result of post-depositional processes. Note that residues can also appear random or uncharacteristic when a tool has been reshaped or resharpened.

Most of the SBL artefacts contain polish on all the raised surfaces and sides, such the dorsal ribs, the bulbar area and the edges. This polish is not always use-related. Use-related polish is often associated with micro fracturing and is confined to used surfaces and raised areas, which were subject to friction, for example the bases of hafted points show polish. When polish is present on every ridge, raised surface and edge, it may originate from recent rubbing with, for example, the plastic storage bag. The distribution maps in Appendix D only indicate polish that can result from

usage. I mainly found use-related polish along possible working edges and along the base of points

In this chapter I discuss the association between different types of residues. Residues are associated when they share the same or a complementary distribution pattern and when they are in the same location. For example, when fat and bone are both present along a tool edge and when connective fibres (deriving from connective tissue) are present in several fat smears, then fat, bone and connective fibres are associated.

Pièces esquillées (scaled pieces) are tools that have two crushed ends. These ends show step fractures, abrasion and polish. Other flakes in the SBL sample also often show damage and this is indicated in the distribution maps in Appendix D. In these maps, "broken" means that part of the artefact snapped off. "Damaged" means that use wear damage is present; this generally consists of abrasion, some micro fracturing and sometimes polishes. The severity of the edge damage is discussed in the text.

SBL1 (Figures D.2 and D.3)

Polish on the surviving piece of the broken proximal end of this hornfels *pièce esquillé* is probably use-related (Figure D.2). I found micro fracturing along the distal end and one lateral side. Fat is abundant on this tool. Most of this fat appears on top of the sediment and is therefore not use-related (Figure D.4), but may obscure other use-related residues. In addition, the distribution maps of fat show no clear or repetitive pattern. However, the ochre and fat deposit close to the broken proximal end is probably use-related (Figure D.5). The polish indicates that this end was subjected to friction and this possibly caused the flake to break. The ochre and fat are perhaps associated. Lombard (2005, 2006a, 2007) found that ochre and plant resin are used in hafting at Sibudu, and that fat might also have been included in the adhesive recipes. Consequently, the tool could have been hafted, and the distal end and lateral sides are the working edges. However, because the tool is broken and because there are no clear residues along the working edges, I cannot say what it was used for.

SBL2 (Figures D.6 and D.7)

Charcoal or burnt plant tissue is copious on this quartz flake. The larger of these deposits is indicated in the distribution maps. Fat is also abundant on SBL2, but it is

mostly found on top of sediment (Figure D.9) and is therefore discounted as a use-related residue. On the ventral side, fat is found next to muscle tissue implying a use-related origin, but on the dorsal side fat is randomly spread over the surface and therefore may not be use-related. Abundant muscle tissue is present and is mainly distributed along the proximal and medial parts of this flake (Figures D.8 and D.10). A few burnt bone deposits are associated with the muscle tissue. The plant fibres are blue and quite bright and they are interpreted as modern contaminants. There is no edge damage on this flake. The muscle tissue and some of the fat and burnt bone deposits are use-related and this flake could have been used to work soft animal tissue.

SBL3 (Figures D.11 and D.12)

This small quartz flake contains abundant fat deposits distributed randomly (Figure D.15), so this could be post-depositional residue. However, additional animal residues such as fatty bone and muscle tissue are also present (Figures D.14 and D.13 respectively) and these support an interpretation of use. Bone and muscle tissue are confined to the distal and medial edges of the flake, but they are small, thin deposits. SBL3 was probably used to work soft animal tissue.

SBL4 (Figures D.16 and D.17)

The distal end of this small dolerite flake is broken. The ventral surface contains several unburnt bone and fat deposits and some fat is present on the dorsal side. The bone is located along the flake edges and on its broken distal end. Because the fat deposits co-occur with the bone along the flake edges, they are probably use-related residues (Figure D.18). The dorsal side of the flake has no visible bone, but it has some fat along the edges. The small and confined deposits of ochre along the dorsal ridge are interesting (Figure D.20). Along this ridge I also found continuous polish, indicating wear (Figure D.19). The ochre and polish seem to be associated. SBL4 shows a clear distribution pattern: fat and fatty bone along the edges and polish and ochre along the dorsal ridge. The fatty bone, fat and ochre residues are all use-related residues and indicate that animal material was processed with this tool.

SBL5 (Figures D.21 and D.22)

This large pointed dolerite flake presents many fatty deposits, which appear most prominently on the distal and medial parts, in association with burnt bone deposits

(Figure D.23). Although some fat deposits occur on top of sediment, at least half are ancient use-related residues (Figure D.27). On the proximal and medial parts of the flake resin, plant tissue and plant fibres are more abundant (Figures D.24 and D.25). The distribution is therefore as follows: animal residues on the distal/medial part and plant residues on the proximal/medial part. Perhaps this flake was used as a lithic component of a spearhead, in which case the plant deposits on the proximal and medial parts derived from hafting, while the fat and burnt bone residues originated from contact with an animal, but no muscle tissue is present on this tool. There are many rootlets on this flake and they are post-depositional (Figure D.26).

SBL6 (Figures D.28 and D.29)

This small dolerite flake contains a multitude of both plant and animal residues. Fatty bone, fat, muscle tissue and plant tissue appear mainly on the distal and medial parts (Figure D.30). The remains are all small and isolated and there are no straightforward connections between residues. The stain on the ventral side is the remainder of some kind of liquid (Figure D.31). Because the fluid soaked into the stone it is impossible to determine its origin. Although all remains are probably ancient, the pattern is difficult to unravel, and the most parsimonious interpretation is that the residues do not derive from use and could be coincidental or post-depositional.

SLB7 (Figures D.32 and D.33)

This tiny dolerite flake contains three minute ochre deposits on the ventral side (Figure D.34). Because they do not occur with other residues and because they are so small and contained, I think this ochre is a post-depositional residue. There are many randomly distributed fat and plant tissue deposits on both the ventral and dorsal sides. Besides one isolated bone flake, the fat cannot be linked to other animal type residues. In addition, the unburnt plant residues are extremely birefringent and bright white (Figure D.35) and I suspect that both animal and plant residues are post-depositional residues and do not derive from use.

SBL8 (Figures D.36 and D.37)

SBL8 was completely covered in a thick and almost compact sediment layer, which is unusual for the SBL sample. This leads me to believe that the tool was used and that the resulting damp residues served as an adhesive for the sediment. This large dolerite

flake is broken along the proximal, distal and one lateral side. The other lateral side shows recent damage. Polish is present along both laterals, but because the flake is so damaged these laterals cannot confidently be assigned to a use. However, the distal tip is relatively undamaged and the polish there could originate from use. Two muscle tissue deposits are present; one along the slightly damaged edge on the dorsal side and the other on the distal tip on the ventral side (Figure D.38). In association with this tissue I found some fat and a connective fibre. Also on the ventral side are two ochre spots (Figure D.39). They are larger than the spots on SBL7, but also appear isolated from other residues. I consider that the muscle tissue deposits are probably use-related because they are rather large, they are associated with other animal residues and they are distributed along a possible working edge. However, I am unfamiliar with the breaking patterns of this type of tool and I cannot assign a precise action to it. The two ochre spots are probably ancient, but because they are so isolated and because the tool is so damaged I am unable to say anything about their origin.

On the ventral side I identified what I labelled a "white starchy" deposit in Chapter Two (Figure D.40). This is a term that has also been used by Williamson (2004, pers. comm. 2004) to refer to Sibudu residues from other periods in the sequence. Similar deposits are present on tools SBL17, SBL23, SBL24, SBL25 and SBL26. Under high magnifications of 500 to 800 times it is clear that this residue does not consist of starch, but of small granules. "White starchy deposits" appear extremely similar to starch residues and can easily be mistaken for one under lower magnifications. However, "white starchy" residues do not contain starch grains. Under high magnifications the white granules show no extinction cross and they remain dull and opaque in (cross) polarised light; this is uncharacteristic for starch granules. It is likely this type has been mistaken for a starchy residue in the past. In the SBL sample many of these deposits lie on top of the sediment, typical for contaminants. Because "white starchy" residues cannot be identified as yet, they are left aside in this study.

Many reflective plant remains, similar to the ones on SBL7, are also present. They are evenly distributed over the surface and appear almost always on top of sediment patches implying that they are post-depositional residues, possibly originating from modern rootlet growth in the sediment.

SBL9 (Figures D.41 and D.42)

The proximal and distal ends of SBL9 are crushed. This small quartz piece is a *pièce esquillé* and the ventral and dorsal sides both contain clear muscle tissue deposits (Figure D.43). They range in colour from dark yellow to dark brown. On the dorsal side muscle tissue is concentrated on the proximal end. On the ventral side thick muscle tissue deposits are also present along one lateral side. On one of the dorsal ridges, clear drops of resin occur (Figure D.44). Some fat is present on the ventral side and charcoal occurs on both sides. On the proximal edge of the ventral side I located a red, fat-like deposit; similar to the fat-like deposits I found on the Sterkfontein archaeological sample and on SBL24 (Figure D.122). It is unclear what this deposit is, but due to its bright colour I believe that this may be a post-excavation contaminant. The muscle tissue is a use-related residue. It is abundant and associated with the working edges. The resin drops are also ancient, but the fat and charcoal could be contaminants. Charcoal could have attached to the tool because of proximity to ancient hearths, which are abundant at the site. Fat is abundantly present on blank samples and on SBL9 it is not clearly associated with the animal tissue or the worked edge. Although I am unsure if the resin drops are use-related, SBL9 was clearly used on animal material.

SBL10 (Figures D.45 and D.46)

This hornfels *pièce esquillé* contains one resin deposit. Because there is no connection to the other residues, and because there is only one deposit, the single resin drop is regarded as post-depositional and part of the "background residues" (Figure D.50). No other plant residues occur on this tool. There are, however, rootlet prints on this piece (D.52). All references to rootlets on the distribution maps are negative prints from disintegrated rootlets. Rootlets are abundant on the lateral sides. SBL10 contained the false "blood residue", which actually is a mark from post-excavational handling (Figures D.47, D.48 and D.49 and see Chapter Two).

SBL10 contains several fat deposits, most of which seem to be use-related. Fat is mainly present along the sides rather than in the centre of the tool. Fatty bone and burnt bone are also present, but there is no clear association between the fat, bone and burnt bone residues. Fatty bone and fat are both located near the lateral edges. Muscle tissue is abundant on one lateral side. Although not found in close vicinity of each other the fat, muscle tissue and fatty bone deposits are associated. They are all animal

tissues and they have a similar distribution pattern along the lateral sides. There are several areas with abrasion and polish (Figure D.51). Some of these abraded areas are located near the edges and some more in the interior of the tool. The animal type residues and the extensive damage, including step fractures and large micro fractures, on the distal and proximal ends indicate that SBL10 was used as a tool to work animal material. While it is unclear what the precise function of this tool is, it involved strenuous action.

SBL11 (Figures D.53 and D.54)

The distal end of this quartz flake shows micro fracturing. Many fat deposits are present and most seem to be ancient (Figure D.55). However, the distribution pattern of fat is entirely random. The plant deposits on the dorsal side are similar to the plant residue on SBL16 (Figure D.79); their isolated position on a dorsal ridge is probably not use-related. These plant deposits are almost certainly part of the sediment matrix. On the ventral side, there are several fatty bone and burnt bone deposits (Figure D.57). Fatty bone is concentrated on the proximal and medial parts. Muscle tissue occurs along the thinner edges of the lateral and distal sides. The muscle tissue deposit in Figure D.56 occurs parallel to the lateral edge, perhaps indicating a cutting motion. The fat, muscle tissue and fatty bone are probably use-related residues and indicate that SBL11 was used to process animal material.

SBL12 (Figures D.58 and D.59)

This small quartz *pièce esquillé* is damaged along all sides. On the ventral side only one small resin deposit is present on the edge. Because it is isolated, and the only residue of its kind, it is discounted. The dorsal side contains a few burnt bone, fat and muscle tissue deposits. The fat and muscle tissue are found relatively close together on the proximal side (Figures D.60 and D.61). The burnt bone occurs close to the distal edge. Because the fat and muscle tissue are associated and because the distribution along an edge makes sense, the residues are considered ancient and use-related. The burnt bone may have come from the Sibudu deposit, which is rich in bone flakes; nevertheless SBL12 has certainly been utilised on animal material.

SBL13 (Figures D.62 and D.63)

On the edge of the dorsal side of this large pointed dolerite flake, fat, fatty bone and muscle tissue are found close together and they are clearly associated. Muscle tissue also appears in small deposits along the edges of the flake, in two larger deposits on the proximal end of the ventral side (Figure D.65) and on the distal tip of the dorsal side (Figure D.66). Many random fat deposits are present and, although they could be ancient, their distribution is difficult to interpret. The plant tissue on the ventral side is similar to rootlet remains on SBL18, SBL19, SBL22 and SBL30 (Figure D.64). In addition, the rootlet is isolated and does not appear to be associated with the animal residues. In conclusion, the muscle tissue, fatty bone and some of fat residues are ancient, but the distribution pattern cannot be interpreted.

SBL14 (Figures D.67 and D.68)

This small hornfels *pièce esquillé* has a broken distal end and the opposite side is damaged with step fractures and some crushing. There are many fat deposits spread randomly over the surface. Most of these fat deposits are ancient and ash overlies the fat (Figure D.69). Furthermore, the tool contains some burnt bone remains (Figure D.70), which also appear random. Although the fat and burnt bone residues are use-related, their distribution pattern is not diagnostic. Close to the broken edge on the dorsal side there are fatty bone flakes (Figure D.71) and some muscle tissue. Another small muscle tissue deposit is present on the proximal end. Although animal products are clearly associated with SBL14, I cannot assign a function to this tool.

SBL15 (Figures D.72 and D.73)

This dolerite flake is broken at the distal and proximal ends. There is no clear residue pattern; this is probably partly caused by the damage to the surface. The (burnt) bone, fat, muscle tissue and plant tissue residues are spread randomly over the surface and they do not appear to be associated. Half of the fat deposits seem to be post-depositional contaminants (Figure D.75). The two fragmented pieces of plant tissue on the dorsal side are similar to the rootlet on the same side (Figure D.74) and to rootlets in SBL13, SBL18, SBL19, SBL22 and SBL30. The isolated single ochre spot in Figure D.76 is probably not use-related. It is unlikely that any of the remains are use-related; however they could be ancient, but resulting from non-use-related handling.

SBL16 (Figures D.77 and D.78)

This small broken hornfels flake contains few micro remains. Only one fat deposit (Figure D.80) lies beneath the sediment on the tool surface and this residue may be use-related. The reflective deposit on the ventral side probably consists of plant tissue, but it is clearly part of the sediment matrix (Figure D.79). The only other plant residue is a modern fibre on top of the sediment (Figure D.81). This flake was probably unused.

SBL17 (Figs D.82 and D.83)

This artefact is a hornfels flake. The plant residues on SBL17 resemble each other and are concentrated around the sharp edge, which shows signs of use. The few plant residues that are located closer to the obtuse proximal side could result from use. I found similar patterns in the experimental sample. Large deposits of muscle tissue are also present along the working edge (Figure D.84). The striations in the deposit on the distal edge are oriented parallel to the working edge and they imply a cutting movement. A dark area on the distal end is visible with the naked eye. However, under the microscope, the stained surface in this area is similar to the surrounding lighter hornfels. It is likely that fat or some other liquid was absorbed into the stone in this area. I am also unable identify other similar small stains on the dorsal side (Figures D.85 and D.86). These stains occur in isolated, small and randomly dispersed deposits. Although the combination with other animal type residues such as the muscle tissue and the one connective fibre appears logical, there is no obvious association and these stains are probably a post-depositional contaminant. The isolated “white starchy deposit” lies on top of sediment and is post-depositional (Figure D.87); this type of residue is discussed in more detail for SBL8.

Because of their quantity and association with the working edge, the plant and muscle tissue remains and the unknown stains are probably use-related residues. However, I am unable to interpret the combination of plant and animal remains and therefore the tool function remains unclear.

SBL18 (Figured D.88 and D.89)

This pointed dolerite flake has a broken tip. There are several deposits with rootlet remains (Figures D.90 and D.91). Other plant tissue deposits are found in the vicinity of these rootlets and have a random distribution over the surface. It is therefore

unclear if the plant remains are use-related residues or, like the rootlets, post-depositional residues. Because the single burnt bone deposit on the ventral side is isolated, it is interpreted as a post-depositional residue. SBL18 also contained several fat deposits, but they were all located on top of sediment deposits and were thus regarded as post-depositional residues. This flake is almost certainly unused.

SBL19 (Figures D.92 and D.93)

SBL19 is a broken dolerite flake. The charcoal, present in the sediment, and the rootlet remains (Figure D.94) are clearly post-depositional. The burnt bone is sometimes present on top of sediment deposits and in addition the distribution appears random (Figure D.95). The small fatty bone and muscle tissue deposits on the dorsal side are located near a possible working edge and could be use-related. However, they appear isolated and not part of a pattern. SBL19 may not contain use-related residues.

SBL20 (Figures D.96 and D.97)

SBL20 is a large dolerite flake. Fat is distributed randomly over the surface, but the residues on the distal and proximal ends of the ventral side appear use-related. The use-related fat residues are quite large and are found in combination with fatty bone and connective fibres (Figure D.98). The other fat remains are small, blue and white deposits and are dissimilar from these larger yellow and white use-related residues. Several of the small fat deposits also appear on top of sediment. The dorsal side contains many contaminants, including one large rootlet, which is visible with the naked eye. Other contaminants are insect remains, charcoal, plant fibres and several small fat deposits. Figures D.99 and D.100 illustrate the unidentified “brown and white deposit”. Because the white part contains small starch grains, this deposit probably has a plant origin. This deposit can be deteriorating resin; it appears to be similar to what Lombard (2006a, 2008) found on other Sibudu tools and experimental tools hafted with *Acacia* gum as an adhesive. This residue needs further investigation and although I found similar deposits on SBL22 and SBL29 (Figures D.108 and D.148), for the time being it remains is unclear exactly what this residue is and therefore it is disregarded.

The largest of the genuine fat deposits is present on the obtuse proximal end and the only other associated residue is located on the distal tip. The distribution appears random and the residues are probably not use-related.

SBL21 (Figures D.101 and D.102)

The proximal edge of this small quartz is damaged and shows small step fractures and some abrasion. Muscle tissue is present on the distal and medial parts and not obviously associated with the proximal working edge (Figure D.105). Fat, although not found under sediment, has a random distribution and is not directly associated with the damaged edge or the muscle tissue. SBL21 is extremely small and this complicates the analysis of distribution patterns; it is difficult to deduce if fat and muscle tissue are associated or not. The unknown dried liquid deposit could be a plant exudate (Figures D.103 and D.104), but it is more likely to be blood plasma, considering the other animal residues. Because there are many relatively large fat and muscle tissue residues, which may be associated, muscle tissue and fat are considered use-related. Unfortunately, SBL21 is broken and the origin of the dried liquid remains uncertain, but considering the nature of the remaining residues SBL21 was possibly used on animal tissue.

SBL22 (Figures D.106 and D.107)

SBL22 is a large and bulky dolerite flake. The ventral side contains an unknown "brown and white deposit", which does not contain small starch grains under high magnifications (Figure D.108). It is therefore unclear if the "brown and white deposit" is deteriorating resin as proposed for a similar deposit on SBL20. Both SBL20 and SBL29 contain this brown and white deposit. Because the deposits appear to be deliberately smeared onto the surface, both Lombard (pers. comm. 2007) and I believe this residue is use-related; however we cannot identify it as yet and it is left aside in this study.

Rootlets, which are abundant on SBL22, complicate the identification of the other plant tissue deposits (Figures D.109 and D.110). It is unclear if other plant remains are rootlet parts or genuine residues. All plant remains are similar; they show large cellular structures and are extremely birefringent. As a result, the plant residues are disregarded because they are ambiguous. The burnt bone is randomly dispersed over the surface and it is unclear if the burnt bone is associated with the also randomly spread unburned fatty bone. The remaining fat, muscle tissue and fatty bone are probably use-related residues (Figure D.111). The combination of these three types is sound, but the distribution patterns appear random. On the dorsal side fat, fatty bone and muscle tissue occur once on the same lateral edge, but in all the other cases the

residues are found away from the thin edges, towards the obtuse and thick centre of the flake. These animal residues are probably use-related and perhaps the flake was used to cut animal tissue. The residues away from the edge could be the result from handling.

SBL23 (Figures D.112 and D.113)

On this dolerite *pièce esquillé* most fat deposits occur on top of sediment patches, compromising other fat remains (Figure D.115). Bone and fatty bone is also present on top of sediment and so is burnt bone. The red deposit in Figure D.114 is not use-related red ochre. This is implied by its position on top of a sediment patch and the lack of clear granules. There are also several "white starchy" deposits on this tool (Figures D.116 and D.117). Similar deposits are present on SBL8, SBL17, SBL24, SBL25 and SBL26. Because "white starchy" residues cannot be identified as yet, they are left aside in this study. See SBL8 for more details on this residue type.

This leaves a few ambiguous fat residues and one small deposit of muscle tissue. These residues are not clearly associated with the damaged edge or each other and the pattern, which emerges is similar to what could be expected from unintentional handling. The edge damage indicates that SBL23 was probably used, but the residues cannot clarify what material was processed.

SBL24 (Figures D.118 and D.119)

This dolerite flake contains many "circular plant deposits". As illustrated in Figures D.120, D.121, D.123 and D.125 these accumulations consist of small pieces of yellow to dark brown birefringent plant pieces, deposited in a circle. The deposit in Figure D.125 is associated with resin and this confirms the plant nature of this deposit. It is unclear what these deposits represent and as such I am unable to draw any conclusions about the authenticity of these residues. In addition, the fat residues, the white starchy deposits (Figure D.124) and the red fat-like deposit (Figure D.122) are all post-depositional contaminants. No remains appear to be associated with the probable working edge and SBL24 was probably unused.

SBL25 (Figures D.126 and D.127)

The distal edge is slightly abraded. There are many muscle tissue and fat deposits on this tool (Figure D.128). They are associated with each other and the possible working

edge. The animal deposits on the proximal end can result from handling during the action. I found a similar distribution of use-related residues in my experiments. The white starchy deposit is a contaminant. Bone and burnt bone remains are found on top of sediment or away from other residues and the working edge and therefore they are probably post-depositional remains (Figure D.129). SBL25 was probably used to work soft animal tissue.

SBL26 (Figures D.130 and D.131)

SBL26 is a small hornfels *pièce esquillé*. The (burnt) bone (Figure D.133) and fat remains are small and isolated from each other. They are also randomly dispersed and do not appear to be associated. The large plant residues on the ventral side appear to be smeared onto the surface and the two deposits are identical (Figure D.132). However, the association between the plant tissue and the fat and bone is unclear. The white starchy residue is a contaminant (Figure D.134). Because there is no apparent meaningful distribution pattern and no association with other residues, the plant tissue is probably an unintentional residue. The animal type residues are ambiguous.

SBL27 (Figures D.135 and D.136)

This extremely small quartz *pièce esquillé* contains few residues; the ventral side shows no remains. The yellow "deposit" on the dorsal side, which is visible with the naked eye, is actually an inclusion (Figure D.138). This is illustrated in Figure D.139, where the yellow inclusion is clearly part of the rock and not a layer on top of it. The small muscle tissue deposits on the dorsal ridge (Figure D.137) appear ancient, but because they are the only remains and because there is no meaningful distribution pattern, I am unable to interpret them.

SBL28 (Figures D.140 and D.141)

This burnt dolerite flake has a broken distal end and SBL28 contains several areas with use polish. SBL28 was covered in thick sediment; after brushing I found brush marks on the surface (Figure D.142). It is possible that light residues, such as starch grains or loose fibres, have been displaced, but residues like fat and resin, are still attached to the surface and their position on the tool is reliable. The large red deposit in the dorsal side is a mixture of ochre with either resin or fat. Compare Figure D.143 with the acacia gum/resin, beeswax and ochre mix in Figure A.22. This residue and its

location close to the proximal edge and polish areas can be indicative of hafting (Lombard 2006a, Lombard 2006b, Lombard 2007) (Figure D.145). Resin is present on the ventral and dorsal side. SBL28 contains many fatty bone deposits along the edges (Figure D.144). The plant deposit on the dorsal side is similar to the plant residues on SBL26. The residues described above appear genuine; they are located under sediment and the tool does not appear to be particularly contaminated with other remains besides insects or rootlets. In addition, residues unintentionally functioning as an adhesive are probably responsible for the thick sediment layer that covered the tool initially. Lombard (pers. comm. 2007, and also see Lombard 2008) suggests that SBL28 could have been hafted in a bone shaft. This explains the combination of resin, ochre, fat and bone and the presence of polish. The plant residue can come from twine also used in hafting.

SBL29 (D.146 and D.147)

The distal tip of this dolerite flake is broken. SBL29, like SBL20 and SBL22, contains a "brown and white deposit" (Figure D.148). Although this residue is considered ancient, it is excluded from this analysis because I cannot identify it.

On the ventral and dorsal side fat (Figure D.148), bone and muscle tissue are present. These deposits are mainly concentrated along the thin edge of one lateral side; left lateral edge on the dorsal side. However, one plant tissue and one resin deposit also occur on the ventral side (Figure D.149). The single isolated resin and plant residues are ancient, but unintentional. The animal tissues present a meaningful pattern and are use-related. Because the distal tip is broken I cannot reconstruct the precise function of SBL18, but it was probably used to process animal material.

SBL30 (Figures D.150 and D.151)

This dolerite flake contained thick sediment deposits, which are associated with use. The numerous rootlets, mainly present on the ventral side, are visible with the naked eye (Figures D.152, D.153 and D.154). Around several rootlets I also found plant tissue deposits. Due to their association with the rootlets I discarded the plant tissue. There are various ancient animal deposits found away from the thin possible working edge (Figure D.155). Fat is present in small smears and on the dorsal side, fat is associated with muscle tissue. Bone is not abundant and only occurs twice. The distribution of these remains appears random and there is no preference for any edge.

The flake may have been used to slice muscle tissue. This action leaves almost no use wear and meaningful residues could be located away from the working edge. The one large animal deposit on the obtuse and proximal end could be the result from handling during cutting. Because the flake is clearly contaminated and disturbed by rootlets I am hesitant to give a definite conclusion.

Table 6.1: Overview of the results from the Sibudu samples; ?= ancient residues, which may be use-related, but unable to interpret the pattern; --= No ancient or use-related residues are present; No= residues are ancient, but not use-related.

Tool Number	Stone	Residues	Interpretation
SBL1	*Pièce esquillé*	Fat and ochre	?
SBL2	Flake	Burnt bone, muscle tissue, fat	Work soft animal tissue
SBL3	Flake	Fatty bone, muscle tissue	No
SBL4	Flake, broken	Fatty bone, fat, ochre	Process animal tissue
SBL5	Flake	Fatty bone, fat plant tissue, plant fibres, resin	Hafted?
SBL6	Flake	Fatty bone, fat, muscle tissue, plant tissue	No
SBL7	Flake	--	--
SBL8	Flake, broken	Fatty bone, fat, muscle tissue, ochre	?
SBL9	*Pièce esquillé*	Muscle tissue, resin	Work animal tissue
SBL10	*Pièce esquillé*	Fatty bone, fat, muscle tissue	Work animal tissue -strenuous-
SBL11	Flake	Fatty bone, muscle tissue, fat	?
SBL12	*Pièce esquillé*	Muscle tissue, fat	Work animal material
SBL13	Pointed flake	Fatty bone, muscle tissue, fat	Work animal material
SBL14	*Pièce esquillé*, broken	Fatty bone, muscle tissue, fat	Work animal material?
SBL15	Flake, broken	Fatty bone, muscle tissue, fat, plant tissue, ochre	No
SBL16	Flake, broken	Fat	No
SBL17	Flake	Fatty bone, muscle tissue, plant tissue	Cut animal tissue?
SBL18	Pointed flake (tip broken)	--	--
SBL19	Flake, broken	Fatty bone, muscle tissue	No
SBL20	Flake	Fatty bone, fat	No
SBL21	Flake	Muscle tissue, fat	Scraper? On animal tissue
SBL22	Flake	Fatty bone, muscle tissue, fat	No
SBL23	*Pièce esquillé*	Muscle tissue, fat	No
SBL24	Flake	--	--
SBL25	Flake	Muscle tissue, fat	Work soft animal tissue
SBL26	*Pièce esquillé*	Plant tissue	No
SBL27	*Pièce esquillé*	Muscle tissue	No
SBL28	Flake, broken	Fatty bone, fat, resin, plant tissue ochre	Hafted in bone shaft?
SBL29	Flake, broken	Fatty bone, muscle tissue, fat, resin, plant tissue	Work animal tissue?
SBL30	Flake	Fatty bone, muscle tissue, fat	?

6.3 DISCUSSION

The study of this 30-tool assemblage reconfirms that organic residues preserve on stone tools from Sibudu Cave (also see Lombard 2004, Williamson 2004, Lombard 2005, Williamson 2005, Lombard 2006a, Lombard 2006b, Lombard 2008). On 27 tools I found use-related residues and in 12 cases I could confidently interpret the processed materials. Only three tools did not contain any residues. The majority of the ancient residues are animal types and these included fat, fatty bone and muscle tissue. The residues discussed in Table 6.1 are ancient because:

- They are not present in the sediment samples
- They occur under sediment patches on the tool's surface
- They are not associated with obvious contaminants, such as insect remains or rootlets

Some residues are use-related because, in addition to the traits above:

- They are present in larger quantities, meaning that single residues could be coincidental, but several associated deposits provide a stronger case for being use-related.
- Their distribution pattern is coherent, often meaning in association with a possible working edge
- They are associated with other genuine residues and this can be in the form of:
    - Similar residue types, for example fat and muscle tissue are animal type residues
    - Sharing a similar or complementary coherent distribution pattern
    - Location close to each other, for example fatty bone flakes in a fat smear

Animal type residues, including muscle tissue, fatty bone and fat, are abundant in the SBL sample. Plant residues are present in the form of plant tissue and resin. In addition I found ochre, an inorganic residue. Four out of the eight *pièces esquillées* have an obvious animal signal; the remaining four contain no residues or what I have interpreted as unintentional residues. The latter is remarkable because the pieces are extensively damaged along the edges and they were almost certainly used. The residues interpreted as use-related on the scaled pieces are not found along the

damaged edges, but on the lateral edges. Further study of a larger sample of *pièces esquillées* may clarify this issue.

The large frequencies of animal residues on these tools are partly influenced by the fat and burnt bone deposits. Parts of the fat and burnt bone remains are use-related, as discussed in the Results section. However, their universal presence on all the samples is indicative of residues that have accumulated by some route that did not involve processing of animal products. Therefore these residue types are not diagnostic and this makes the interpretation of fat and burnt bone that is spread randomly all over a tool difficult. Some bone possibly burnt after a tool was disposed in or around a hearth (also see Chapter Two), in which case the burnt bone can be use-related. In order to understand the integrity of residues on a tool, it is important to understand where contaminants originate. Burnt bone is present in the sediment matrix. It is also unclear how much fat is unintentionally left on a tool's surface after handling during ancient times. More study and experimental work on fat and burnt bone is greatly needed. For now, I will leave these results as they are; obvious contaminants are disregarded and because I used a contextual approach, based on association and distribution, following Lombard and Wadley (Lombard and Wadley 2007, Wadley and Lombard 2007, also see Chapter Two), single and incoherent residues are also dismissed. The interpretation of tool use depends on the full spectrum of residues and their distribution; therefore weaker strands of evidence become less significant (Lombard pers. comm. 2007).

Muscle tissue is abundantly present on the SBL tools. Perhaps this is because of some morphological trait such as birefringence, which could have been previously mistaken for a plant residue in other studies (Wadley, Lombard and Williamson 2004, Lombard and Wadley 2007, Wadley and Lombard 2007). It is also possible that muscle tissue is overrepresented in this SBL sample. Muscle tissue is extremely obvious on quartz tools due to the reflective stone surface. Even thin and small deposits are unmistakable. On dolerite, hornfels and other dark stone types from Sibudu, thin muscle tissue deposits are less visible and it is more likely that muscle tissue is underrepresented on dark rocks. As discussed in Chapter Four, thick hyphae deposits can be mistaken for muscle tissue. However, on the SBL sample almost no hyphae are present; spores are also rare. It is unlikely that muscle tissue has been misidentified as hyphae. From the experiments in Chapter Four it is apparent that

muscle tissue is fragile and generally decays rapidly. Muscle tissue preserves best in a dry environment, like Sibudu, but it is unclear why there is so much muscle tissue preserved on the ancient Sibudu tools. Future research should clarify this issue.

The Sibudu sediments also contain burnt bone and the blank sample shows non-use-related fat residues. However, I have not found any remains that are similar and that could mimic muscle tissue in the sediment samples or blanks. Because Sibudu Cave is extremely dry, I predicted in Chapter Two that muscle tissue would preserve inside the shelter. However, I also predicted that the other residue types, plant tissue, starch, resin, fat, blood and bone would preserve. Although it is extraordinary to find so many muscle tissue residues, I suggest that these residues are accepted as muscle tissue remains until more research is done on this residue type.

Lombard (*e.g.* 2006b, 2008) and Williamson (2004, 2005) also found animal type and plant type residues and several inorganic types such as ochre and vivianite. Lombard and Williamson independently analysed large Sibudu samples, which were carefully collected and curated for this purpose. The tools were collected during the excavation and immediately stored in plastic bags. The samples were not handled before the residue analysis. Both Lombard and Williamson analysed sediment samples and extensively photographed their findings. Lombard conducted meticulous analyses of the distribution and association of residues (Lombard 2004, Lombard 2005, Lombard 2007, Lombard 2008). She was able to show that points from Sibudu were hafted with an ochre resin mix and that some Howiesons Poort pieces were hafted in bone shafts.

The Results section reveals several complicated residues. One residue type is the "white starchy deposit". This deposit does not contain starch grains, but this is only apparent when the residue is analysed under high magnifications. It is possible that some analysts incorrectly identify similar deposits as a starch deposit. Due to space and money constraints many publications do not contain (many or colour) pictures of residues. Therefore it is not possible to make comparisons with other residue studies. I am unsure what this white granular deposit is, but it could be ash. More studies and experimental work are needed.

The "blood residue" on SBL10 presents a cautionary tale. The cracked stone surface appears strikingly similar to the mud cracking, which is characteristic of dried

blood. The mistaken identity only became apparent under high magnifications and after additional study of the "residue".

The "brown and white deposit", discussed in the Results section under SBL20 and SBL22, these deposits are probably ancient. Some deposits contain small starch grains, but others do not. Perhaps this residue type is deteriorating resin, but more work on this type of deposit is needed.

The SBL sample demonstrates that meaningful residue analysis relies on a sound methodology. I am not always sure what a particular residue type is, or whether it is ancient and use-related. Using the contextual approach improves the reliability of residue analysis, but one must always allow a percentage of error (also see Lombard and Wadley 2007).

Table 6.1, which presents the results, illustrates the need for quantification. It sums up the results, but without the distribution maps and the micrographs it is not clear why the identification of certain residues does not lead to an interpretation of processed materials. Unintentional residues are not separated from use-related residues. It is important to somehow quantify results, for example with distribution maps or by counting (*e.g.* Lombard 2004, Lombard 2005, Lombard 2006b, Lombard 2008). Otherwise single ambiguous residues appear to be as important as clusters of indisputable residues.

The results from the SBL also demonstrate that when genuine residues are preserved they are unambiguous. Although it can be difficult to identify a particular residue type and single residues are complicated at times, in general, use-related residues are obvious. This places the archaeological Sterkfontein sample in perspective; this sample only contained ambiguous residues. The Sibudu sample preserves perfect residues, which allow analysts, like Lombard, to answer archaeological key questions. The reason for the preservation is a combination of the sheltered and extremely dry environment that the overhang provides, initially alkaline anthropogenic sediment, and the quick burial of tools. The rapid desiccation of residues ensured preservation. In the shelter microbial activity is low, allowing preservation. In addition the tools were not reworked and the site is in primary context. Furthermore, the majority of the rock consists of dolerite and hornfels and these dark materials allow for easy viewing under the microscope. Hornfels and dolerite are fine- to medium-grained rock types

and they are excellent for residue preservation. Sibudu is an exceptional site for residue analysis. The situation at Sterkfontein is the opposite and there is no reliable archaeological residue preservation at this site.

The archaeological results concur with the experimental results; after one year residues are well preserved inside the Sibudu shelter. The archaeological sample shows that in a stable situation, residues remain intact.

6.4 CONCLUSION

The archaeological sample from Sibudu Cave preserves animal, plant and inorganic type residues well. There is a possible bias in residue identification as there are many muscle tissue remains. The dry environment at Sibudu is ideal for muscle tissue preservation, but because muscle tissue is fragile I had not expected to find so much of it. This point needs more research. There is a clear correlation between the preservation of experimental and archaeological residues; on both samples use-related residues are present.

Sites with little microbial activity, in this case caused by aridity and initial alkalinity, preserve residues better. Sheltered sites, like Sibudu are more likely to preserve residues than unstable open-air sites, like Sterkfontein. Furthermore, stable sites like Sibudu, preserve residues better then sites that underwent post-depositional processes, such as Sterkfontein. The SBL sample is well curated and analysis is therefore straightforward. In contrast, the Sterkfontein residue sample has not been controlled. Therefore residues on the Sterkfontein sample are ambiguous and interpretations are uncertain.

Conditions at Sibudu are optimal for residue analysis. The shelter is dry, the sediment stable and, in addition, the majority of stone tools are made on black fine-grained material, which allows for good viewing and preservation. Sibudu is a unique site from this point of view. Nonetheless, there are contaminants evident on some of the tools, even though the collection was carefully curated specifically for residue analysis. This shows that contamination occurs readily and that excavators and residue analysts alike must make every effort to ensure a pristine collection for analysis. Packaging materials need to be reviewed, for example.

7 SYNTHESIS

In first part of this Chapter I discuss the results and ideas that arose from the previous chapters. In addition, I propose some ideas for future research. In the second half of the discussion I aim to synthesise my results and the resolution of issues that arose in the Introduction.

7.1 DISCUSSION OF THE RESULTS

The predictions, made in Chapter Two, were confirmed in the course of this study. Good preservation of organic residues was expected and was demonstrated inside the modern day Sterkfontein Caves and Sibudu Cave and on the ancient Sibudu >60 ka sample. The remaining experimental and ancient settings were not expected to preserve residues and, indeed, they did not. In the following paragraphs I discuss the results obtained from the experiments and the ancient samples.

Mistaken identity, the blank sample and the contextual approach

Bruier (1976) warns that plant type residues are not always artefactual. Plant remains from the sediment sometimes adhere to tools and Barton *et al.* (1998) agree that great caution should be exercised when interpreting plant residues. However, my experiments show that the identification of animal type remains are just as complicated and should not automatically be interpreted as artefactual.

On the Sterkfontein experimental blanks I found a residue that is similar to fat. Visually it is identical to fat droplets from the replicated sample. I also found a deposit that is similar to muscle tissue. On the Sterkfontein and Sibudu experimental blanks I found fat-like remains. They have a bluish and opaque appearance. On one Sibudu blank I found a fibre, which appears similar to a connective fibre. A few fatty bone-like deposits are also present on the experimental Sibudu sample. On many Sibudu blanks burnt fatty bone deposits are present. It is possible that these fat and bone residues originate from the sediments. In addition, residues that look like animal products can come from handling, which produces finger grease. Single contaminant

residues can be mistaken for use-related residues. Later in this Discussion I explain that, for this reason, analysis of single incoherent residues is useless.

I found that starch grains can deteriorate and become unidentifiable and sometimes even unrecognisable. They become opaque and dull after contact with hyphae and they lose their diagnostic extinction cross and shape. Other plant tissues also change during contact with hyphae. They lose their rigid shapes and cell structures, leaving masses that resemble muscle tissue. In other situations, degraded plant tissue can resemble an ash or starch deposit. Cell structure is lost and only many small fragments remain, sometimes consisting of small starch grains. When no starch grains are present and the particles are more cohesive, this deposit can also resemble bone.

Hyphae can resemble muscle tissue. Hyphae and muscle tissue have no clear cell structures, both materials can be birefringent, and the tissues have a gelatinous constituency and can have holes in their structure. Any residue that is (being) decomposed by hyphae is at risk of being mistaken for muscle tissue.

Contaminant residues adhered to the unused blank flakes. The worked tools contained much more sediment and sediment organics. It is probable that the moist, use-related residues act as adhesive. On the blank sample organic sediment remains are randomly distributed.

Besides plant remains and sediment particles, hyphae were also present as contaminants on blanks and therefore cannot be used to reconstruct tool use as suggested by Haslam (2005).

The blanks and sediment samples provided insight into the range of available sediment contaminants. The blank sample also proved useful because it familiarised me with the way in which residues attach to the rock types that were also used in the past.

It might appear that residue analysis is too complicated to be successful. However, there are solutions for the problems discussed above. The experimental samples demonstrated that use-related residues can be identified by using the contextual approach. Use-related residues are present in suites; these are coherent combinations such as starch and epidermal tissue. Unlike sediment remains, use-related residues have an evocative distribution. On the experimental samples they were present along

the working edges and sometimes on the obtuse ends and sides. Handling caused the latter. Different functions leave different distribution patterns and experimental research into hafting and tool use is important to recognise and interpret residue patterns. Also important in the contextual approach is the quantification of residues. When residues are preserved they are present in larger quantities than contaminant residues. Because it is impossible to make a distinction between use-related and post-depositional residues based on one fragmentary residue, only the contextual approach and suites of residues can reconstruct the processed materials on tools.

The ancient samples

Use-related residues are present on the ancient Sibudu tools. As predicted, the Sterkfontein artefacts did not preserve any residues. The Sibudu residues demonstrated that meaningful patterns can also be discerned on ancient tools as Wadley and Lombard (Wadley and Lombard 2007, Lombard and Wadley 2007,) claim. Moreover, their approach to distribution and association of residues is also useful for distinguishing use-related residues from contaminants.

Contrary to previous research by (Loy 1998a, Loy pers. comm. 2002), the Sterkfontein tools did not yield convincing residues. Perhaps the discrepancy between my results and Loy's positive results can be explained by a difference in methodology. I did not expect residues to be present on the ancient sample because the experimental tools (outside the cave) only preserved a few residues after just one year. Outside the cave biological, chemical and probably also mechanical weathering, caused by microbes, rain, wind and sediment movements, deteriorated most residues. Sterkfontein archaeological deposits are not in primary context and even though experimental residues preserved inside the cave, the ancient tools were originally deposited in the destructive environment outside the cave. Therefore Jones's (1998) predictions that residues should preserve underground at Sterkfontein are irrelevant.

Problems with the Sterkfontein archaeological sample were exacerbated by its complex curation history (explained in Chapter Two). The tools were handled extensively before Loy's analysis (1998a, Loy pers. comm. 2002) or my one, leaving bone-like skin flakes, finger grease, plant fibres, starch grains and other contaminants on the tools. From my analysis it is obvious that residue analysis cannot be applied to samples with such a taphonomic history.

As a further complication, most of the archaeological tools from Sterkfontein were made on quartz. Quartz is highly reflective and difficult to view and this glass-like material only allows residues to preserve in cracks and not on the general surface.

The remains that are present on the ancient tools are also present in the sediment and breccias samples, on the modern blank flakes and on top of sediment patches or on nail varnish used for labels. In addition, the distribution of these remains is random and this is typical for contaminants. If these micro-remains were ancient use-related residues, one would expect to see meaningful residue distribution patterns within, for example, the retouched flake group.

The DNA analysis on ancient Sterkfontein blood samples is unreliable. Williamson (2000b) and Loy (Kuman pers. com. 2007) did not meet the criteria set by Cooper and Poinar (2000) to exclude modern contamination. In addition, analysis on soft tissue is unreliable, the samples were too old and the extraction methods were not optimal. Because the Sterkfontein tools were handled extensively, they should have been excluded from DNA analysis (Ambler and Daniel 1991, Eglinton and Logan 1991, Handt *et al.* 1994, Poinar and Stankiewicz 1999, Hofreiter *et al.* 2001, Leonard *et al.* 2007).

Because the Sibudu experimental sample (inside the shelter) demonstrated good preservation, I expected to find ancient residues. On 27 of the 30 Sibudu tools I found use-related residues and in 12 cases I could confidently identify the processed materials. The archaeological sample demonstrates that in a stable situation, residues remain intact. These use-related remains have meaningful distribution patterns and are present in suites of coherent residues. The results from the Sibudu sample also demonstrate that when residues are preserved they are not ambiguous as the remains on the ancient Sterkfontein sample are. In addition, when residues are preserved, they are present in larger quantities. This again indicates that analysis of single residues is not constructive.

The reason that residues remain preserved at Sibudu lies in a combination of the sheltered and dry environment and the anthropogenic and at least initially alkaline sediment and perhaps the quick burial of the tools. As a result of these factors, microbial activity is low, ensuring preservation. In addition the tools were not reworked and the site is in primary context. Furthermore, most rocks are fine- to

medium-grained dolerite and hornfels. These dark rocks allow easy viewing under the microscope and, because the rocks are granular, residues remain attached. The ancient sample was carefully collected and curated for this study and therefore contamination was minimised. These factors make Sibudu an exceptional site for residue analysis.

Residue preservation

The experiments demonstrate that protective environments in caves and shelters, or on a smaller scale under sandpaper-covered slides, have a positive effect on residue preservation. Because in sheltered circumstances the environment is more stable, cave sites generally preserve more organic remains than open-air sites. The Sterkfontein experiments illustrate this point. Outside the cave, where the environment is unstable, residues decayed rapidly and all residues disappeared within one year. Inside the cave, where the environment is stable, residues preserved well after one year. In addition, cave sites are sheltered from rain and remain drier, which inhibits bioactivity and thus leads to better preservation. Nevertheless, residues can preserve at open-air sites when these sites are stable, meaning that no post-depositional processes disturbed the deposits. In addition, residues can be preserved in desiccated or waterlogged open-air sites if the environment is stable.

The sandpaper slide experiments at Zelhem demonstrate that protected residues underneath tools have a better chance of survival. In addition, some experimental tools deposited on top of the sediment outside Sterkfontein cave showed differential preservation on separate faces. The tool side that faced down in the sediment preserved residues better than the side that faced up. This implies that rain and other elements have a deleterious influence on residue preservation, but that the tool protects the residues on the hidden surface from decay.

The sandpaper slide experiments also demonstrate that residues preserve badly on glass-type surfaces and this has implications for the study of artefacts made on quartz and other glass-like surfaces. Preservation on fine-and medium-grained paper is better than on large-grained sandpaper. A large, irregular surface is best for residue preservation. It is possible that residue preservation is influenced by the pH of the raw material. Future research should investigate this.

Microbial activity is the most important factor causing decomposition. Sediment types, such as clay or ash, are better than organic-rich sediments for preservation of residues,

because when microbial activity is high, most residues are lost. Microbes need water, oxygen and a moderate pH to survive. Moist and permeable sediments therefore ensure decay. Nutrient-rich sediments also have higher bioactivity than nutrient-depleted sediments. Residues survive when water and oxygen are unavailable or when extreme pH values interfere with microbe activity. Sites with low bioactivity, such as Sibudu, have good residue preservation, whereas sites with high bioactivity, such as Wilhelmina Polder, have poor residue preservation.

Few or no residues preserved outside the caves at Sibudu and Sterkfontein and in Wilhelmina Polder and Zelhem. Equilibrium, as discussed in Chapter Three, was not researched in these settings. The equation below predicts that "a" numbers of A react with "b" numbers of B to create "c" numbers of C and "d" numbers of D. At equilibrium as much A and B changes in C and D and the other way around. In the beginning only A and B are present and the reaction is fast, but as A and B are consumed the reaction slows down (Raiswell 2001). Therefore residues deteriorate quickly during initial deposition and later, when equilibrium is reached the amount of residue present stabilises; a prediction also made by Barton (Barton, Torrence and Fullagar 1998).

$$\begin{array}{c} \\ aA + bB \leftrightarrow cC + dD \end{array} \quad \begin{array}{c} \uparrow \text{gas} \\ \\ \end{array}$$

With biological decay of residues, equilibrium is never reached because the by-product carbon dioxide is lost as a gas (Cronyn 1990, Raiswell 2001, Watkinson 2001) and the organic residues are eventually completely consumed.

Biological decay can be extremely slow due to inhibiting factors. With slow decay it appears that residue are permanently preserved. However, chemically and sometimes visually these residues undergo change. Slow decay can take up to tens of thousands of years, provided that the environments remain stable. Changes in the environment can cause these inhibiting factors to change and rapid decay could set in.

Chemical decay could reach equilibrium, but only when the secondary products (C and D in the equation) remain in the environment. In permeable sediments, for example, C and D leach out and decay continues until all residues are consumed. Therefore in an unstable environment residues are more likely to deteriorate rapidly.

Some residue types preserve better than others. Plant tissue and fibres preserve better than any of the other tested residues. The cell walls of plant tissue do not readily decompose and plant tissue contains sugars that decay in a more complicated way than those of starch. Compared to bone-working, wood-working also leaves more residues on a tool surface, increasing the residues' survival chances.

Starch is an extremely vulnerable residue and is often not preserved. Starch grains have no cell walls and they are nature's energy storage cells. The grains are easy and rewarding for micro-organisms to break down.

Muscle tissue also preserves badly. Animal cells do not have cell walls and they are easy to break down. Blood is also an extremely fragile residue.

Bone consists of more mineral components and is more complex to decompose than other animal components. Because of the minerals in bone, the energetic reward values are low for micro-organisms compared to those for starch or muscle tissue. Although bone preserves well, bone working leaves few residues and this decreases the residue's survival chances.

Fat contains great quantities of energy, but because it is a complex molecule, it is also difficult for microbes to break it down. Therefore good fat preservation is generally expected, but because fat can get absorbed in the rock and in the sediment, it is often lost for analysis.

Although some residue types more persistent than others, the environment can also lead to differential preservation. Table 3.12 and Figures 3.2 to 3.9 demonstrate that different residues have different preservation optima.

Because residue types preserve differentially, processed materials can potentially be misrepresented. The experimental starch samples often did not preserve any starch grains, but did contain pieces of epidermal plant tissue. When bone degrades, fat becomes dominant in the remaining residue suite. In these two examples, the processed materials could be mistaken for plant tissue and fat, instead of starchy plant and bone respectively. On archaeological material on which residues are likely to be partly degraded, this bias is potentially even more distinct.

Starch grains are used to identify ancient food plants; every species has a particular shape and size that can be classified (*e.g.* Piperno and Holst 1998, Therin, Fulagar and

Torrence 1999, Piperno *et al.* 2000, Balme and Beck 2002, Parr and Carter 2003, Horrocks *et al.* 2004, Horrocks and Lawlor 2006, Barton 2007, Horrocks, Campbell and Gumbley 2007). However starch grains do not preserve well. In other studies starch grains have been extracted from grindstones and from sediment samples (*e.g.* Ungent, Pozorski and Pozorski 1981, Tuross and Dillehay 1995, Piperno and Holst 1998, Piperno *et al.* 2000, Lentfer, Therin and Torrence 2002, Babot and Apella 2003, Piperno *et al.* 2004). Grindstones may provide a sheltered micro-environment which allows preservation. However, once excavated from the stable environment, decay particularly by fungi, will set in. As a result, the grains are damaged and can lose their unique characteristics. In sediment, starch grains are extremely vulnerable. Starch is also a ubiquitous contaminant and the validity of identifying starch grains in ancient contexts needs to be addressed.

Barton, Torrence and Fullagar (1998) demonstrate that contaminant starch is only present on tools in small quantities when compared to use-related starch. Therefore it is important to know how much starch is present on a tool and starch remains should be quantified.

Although use-related patterning is predictable, residues can also appear random or uncharacteristic when a tool has been reshaped or resharpened. To avoid misinterpretation, large assemblages of similar tools should be analysed.

Identifying use-related residues

The results lead to the following practical heuristic list, which indicates whether residues are use-related. Residues on archaeologically recovered stone tools are likely to be use-related when:

- They are not present in the sediment samples.
- They occur under sediment patches attached to the tool's surface.
- They are not associated with obvious contaminants, such as insect remains or rootlets.

And when, in addition to the conditions listed above:

- They are present in large quantities and several deposits; contaminants are generally single isolated residues.
- Their distribution pattern is logical, for example in association with a possible working edge.

- They are associated with other use-related residues and these can be in the form of:
    - Similar residue types, for example, fat and muscle tissue are both animal product residues
    - Sharing a similar or complementary distribution pattern
    - Location close to each other, for example fatty bone flakes in a fat smear

Future research

Some questions and ideas for future research emerge from the results that I have described. Blood is extensively discussed in the literature (*e.g.* Seeman *et al.*, Loy and Wood 1989, Loy and Dixon 1998, Williamson 2000b, Kooyman *et al.* 2001, Shanks *et al.* 2001, Lombard 2004, Williamson 2004) and one would expect to find many blood residues as a result. However, blood generally preserves badly and, because animals are commonly exsanguinated before processing, few blood residues are produced by, for example, butchering and hide working. One does expect to find blood on hunting weapons. It is unclear why blood is so central in the literature and I wonder if the emphasis on this residue type is justified.

I found that muscle tissue also does not preserve well and that even on the experimental sample inside Sibudu Cave, muscle remains were somewhat damaged. However, on the ancient sample from Sibudu I found many muscle tissue residues. I suggest that these residues are accepted as muscle tissue remains. However, more research needs to be done on this residue type. The number of fat deposits on the ancient Sibudu sample also needs to be addressed. A large number of these residues are not use-related and it is unlikely that so much fat can result from handling. The "brown and white deposits" and the "white starchy deposits" also require further investigation. The nature of these remains is unclear, but they could be use-related residues. The burnt bone on the Sibudu samples also needs more study. It would be useful to know whether these residues are unintentional remains or use-related. Even though the Sibudu tool sample was carefully collected specifically for residue analysis, the sample contained contaminants. Contamination occurs readily and excavators and residue analysts alike must make every effort to ensure a pristine collection for analysis. Packaging materials need to be reviewed. For example, the plastic flakes

found on some of the artefacts may come from the plastic bags in which the samples are stored.
On the ancient and experimental samples from Sterkfontein and Sibudu I found many bone-like deposits and I suggest that these remains are skin flakes. However, this idea needs testing. It is also unclear how much fat is unintentionally left on a tool's surface after handling during ancient and modern times. More study and experimental work on fat is greatly needed.

I discussed chemical and biological weathering above, but future research should investigate mechanical weathering. Unstable environments generally have a negative effect on preservation, but there is no insight into the influence of mechanical weathering on residue analysis. It is likely that mechanical weathering destroys residues, but perhaps it also redistributes residues over the tool's surface and thus compromising the contextual approach.

More research on the influences on preservation of direct burial versus prolonged exposure and on moist/fresh versus dried out residues is needed. This could lead to predictions on preservation from a post-depositional viewpoint. Preservation could be influenced by the season of deposition. For example, if a tool with residues was deposited during the dry season at Sibudu it would quickly become desiccated and thus protected from biological decay. If a tool was deposited during the wet season, the residues would have taken longer to dry out and they would have been prone to deterioration during this time. Rapid burial or prolonged exposure could also influence preservation. Sedimentation rates can also be dependent on seasonality.

In the Sterkfontein exposed experimental sample (outside the cave), the flake side that faced down preserved the most residues. However, it is unclear why the exposed tools outside the Sterkfontein Cave score even higher than the buried tools. If the sediment protects the tools from the elements, one expects the completely buried samples to preserve better than the partially covered ones. Perhaps microbial activity is lower in the top sediment, than in the deeper sediment in which the buried tools were deposited. This point needs further investigation. Because the buried bone tools (inside the shelter) were lost in the Sibudu experiments, it is unclear if exposure or burial is better for residue preservation. It would be interesting to investigate this matter, also because it might say something about favourable seasons for deposition.

The residue types discussed in Chapter Three should be tested further to find their optimal preservation circumstances. There are no equal amounts of information available for all residue types. For example, wood decay is extensively studied (*e.g.* Greaves 1969, Blanchette *et al.* 1988, Blanchette *et al.* 1991, Blanchette, Obst and Timell 1994, Clausen 1996, Blanchette *et al.* 1997, Bjordal, Nilsson and Daniel 1999, Bjordal, Daniel and Nilsson 2000, Blanchette 2000, Bjordal and Nilsson 2002), but specific literature on for example fat, animal tissue and resin preservation is sparse.

As discussed above, particular environments probably ensure preservation of certain residue types. It would be interesting to expand our knowledge of the influence of soil and sediment types. More research can lead to detailed predictions about residue decay per sediment type. This would enable quick assessments to be made about a site's suitability for residue analysis.

7.2 GENERAL DISCUSSION

In the Introduction I discussed the research lacuna in residue analysis, which Grace (1996) and Odell (2001) point out clearly: residue specialists make a direct analogy between the residues they observe and materials that were processed. They appear to be unconcerned about the possibility that data might be missing or that they might be dealing with a palimpsest of residues. As a result, residue studies are often regarded as naive. However, residue analysis is a potentially powerful and scientific tool in lithic analysis and as such it must aim to advance itself. In this study I applied middle-range-theory to understand the impact of depositional processes.

Middle-range-theory tries to observe empirically the processes and principles that lead to the formation of the archaeological record (Binford 1977, Binford 1983, Raab and Goodyear 1984, Schiffer 1988). It is necessary to understand these processes because archaeologists want to be able to discriminate between past human activities and non-anthropogenic processes. For example, archaeologists need to know whether an excavated bone assemblage represents a hyena den or a butchery site.

In the course of this study I demonstrated that it is possible to discriminate between residues from use-related human activities and other processes. Haslam (Haslam 2006a) elegantly shows that small-scale research is valuable in its own right, but this line of analysis should not be perused to avoid complex research issues. I

hope that this study is the first of many large-scale middle-range projects, which allow residue analysis to appropriately deal with its shortcomings.
The experiments presented in Chapter Two and discussed in Chapter Four were successful. Experimental archaeology is a useful tool and should be used to solve research issues for residue analysis. Although molecular analysis of residues, as discussed in the Introduction, is valid in its own right, it should not be left to resolve residue research issues. In addition, molecular analysis itself struggles to extrapolate firm results on archaeological questions (Barnard *et al.* 2007). Residue analysts can resolve a large portion of the issues themselves through microscopy alone. This thesis clearly demonstrates that experimental archaeology and microscopy are important tools, which should be used more regularly to test the methodology of residue analysis. For example, there is need for a large residue comparative database. The database ought to contain descriptions and photographic material of replicated, experimental and ancient residues and it should be accessible to all, perhaps on the World Wide Web. In addition, this database should contain residue distribution maps of many replication studies and help to predict and recognise meaningful residue patterns.

Future residue studies should apply the contextual approach. In this approach the parameters are association and distribution. Association is two-fold: there is an association between residues and an association between residues and their placement on a tool. A distribution map should plot all the residues and their spread over a tool. Other researchers suggested a contextual approach in the past (*e.g.* Loy, Spriggs and Wickler 1992, Hardy and Garufi 1998), but general residue analysis papers do not mention the importance of residue distribution, association or quantification (*e.g.* Fullagar 1998, Odell 2001, Fullagar 2006).

Residues should be quantified. Without weighed results, one starch grain could dominate the processed material and this is obviously unreasonable. In some studies starch grains are quantified by extracting them from the tool' surface (*e.g.* Atchison and Fullagar 1998, Fullagar *et al.* 2006). Because this leads to a loss of context, quantification should preferably be done behind the microscope. Fullagar and Jones (2004) describe the number of utilized edges and surfaces (dorsal, medial etc.) in their study and Lombard (2004) quantifies her residues by measuring the

percentage (of square millimetres) of residues on particular tool parts (for example, the medial lateral edge).

Barton, Torrence and Fullagar (1998) analysed blank artefacts, which they deposited in the sediment to gather starch grains. They set out to investigate whether starch from the sediment adheres to tools and if so, whether the concentrations in the sediment and on the tool are the same. They emphasize that used edges must contain more starch grains than the surrounding sediment (Barton, Torrence and Fullagar 1998, 1233). However they extract the grains by immersing the whole tool in water. There is no control for the amount of analysed sediments (in millimetres) that can be compared to a quantity of tool surface (in square millimetres). In their paper they state: "…it seemed likely that starchy residues … would have spread over the entire artefact" (Barton, Torrence and Fullagar 1998, 1234). In this study they demonstrate that used tools contain more residues than unused artefacts, but they do so without mapping the residues, so the quoted statement is speculative.

Most analysts rightly analyse sediment samples. I suggest that a blank set of replicated artefacts should also be set out at analysed sites and that the sediment organics on these blanks be analysed. In this way, the influence of rock types is also studied.

Archaeologists must give residue analysis the place it deserves within a larger research project and make the specialist part of the team. In practice a specialist is sometimes the last person to analyse the tools, whereas considering contamination issues he should be the first. By making a specialist part of the research team, excavation conditions can be controlled and he can also be part of the fluid progress of research questions. Taking on a specialist in the spur of the moment or just to see "what happens" is pointless. Ideally residue analysis should be conducted with solid research questions in mind. Analysis of rare or museum pieces is limited not only because the pieces can be contaminated, but also because these studies add little knowledge to our understanding of ancient people. Similarly Outram (2008) states that the lack of clear aims in some experimental studies limits the usefulness of these studies. However, these small studies are not without value and sometimes the analysis of rare artefacts leads to fascinating insights. For example, although Hardy *et al.*'s (Hardy *et al.* 2001, Hardy 2004) sample is inadequate to answer questions on tool typology, on the basis of a few tool they demonstrate that Neanderthals used avian and fish resources. These findings changed existing views on Neanderthal

subsistence. In addition, Haslam (2006a) shows that single objects can say something about agency.

Preferably residue analysis should be conducted on a large sample. Unused tools can contain residues and part of the obviouly used Sibudu *pièces esquillées* did not show use-related residues. I can conclude that the *pièces esquillées* were used to process animal material, but I cannot say how they were used exactly. If the sample was larger I perhaps would have been able to say more about these tools. I was able to exclude the blue fat smears and the burnt fatty bone from the interpretation because I had studied a moderately large experimental and ancient sample and knew that these residues are present on almost all artefacts. Only with large samples are specialists able to filter out contaminants and allow a proper assessment of a sample. On the basis of a single piece it is complicated to determine what residues are reoccurring contaminants and which are not. By this I also mean that the number of tools per type must be large. For example, although Hardy *et al.* (2001) analysed 50 tools, they only analysed a few of every type. In order to recognise meaningful distribution patterns and contaminants, they should have analysed 50 artefacts per tool type. Residue analysis is more valuable on a large artefact sample of one tool type.

Although Fullagar (2006) and Hardy, Bolus and Conard (2008) suggest that cleaning does not destroy residues, it at least complicates residue analysis. When cleaned with water some portion of the residues disappear and this is disastrous in the contextual approach. From my brushing experiments I learned that some residues are lost and, more importantly, that part of the loose residues are redistributed over the surface. Consequently, the contextual approach is compromised. Tools are sometimes covered in sediment and need to be cleaned before analysis. I recommend light brushing, because although it redistributes residues, fewer residues are lost than with washing. Handling could add new non-use-related residues and as a result it obscures use-related residues. The archaeological Sterkfontein sample demonstrates how easily a tool is contaminated. Cleaning and handling must be minimised before analysis. It is unwise to work on museum collections or to analyse tools after lithic or micro-wear studies have been completed. Any analysis that is conducted on a handled and cleaned sample should be viewed with suspicion, or at least interpreted cautiously.

A residue specialist should always study the taphonomy of a site and investigate whether the modern-day preservation circumstances are similar to those in the past. For example, the preservation circumstances in Sterkfontein Cave are, as

Jones (1998) demonstrates, favourable. However, it is the situation before deposition in the cave, which caused diagenesis and a proper study of the tool taphonomy would have prevented this sample from being analysed and confusion in the literature could have been avoided.

On the experimental samples, fungus is the main decomposer and it is reasonable to assume that this was the same in the past. Organic remains are protected from fungal decay in dry or desiccated environments, and environments with extreme temperatures and/or pH values. Oxygen-depleted environments, such as those that are waterlogged, also ensure preservation. Obviously there is no straightforward answer to the cause of residue diagenesis. All residue types have their own preservation optimum (see Chapter Three) and these optima can conflict. Consequently, there is always some bias in preservation and residue studies should minimally include a prediction for the success rate of residue preservation per residue type. This would allow other researchers to assess the study. In addition, it would provide a suitable response to Grace (1996) and Odell's (2001) compelling arguments about the possibility of missing data. Residue specialists should carefully assess the site from which their material came and measure for example pH, sediment moisture and temperature. When Table 3.12 and Figures 3.2 to 3.9 are refined, it can then be applied to these sites. The predictions I made based on Table 3.12 concur with the results from the experiments and archaeological samples.

Since tools often contain different residue types and because these cannot be conserved individually, I advise storing promising tools under dry and cold conditions, for example, in well-sealed containers in a refrigerator. The cold conditions and the lack of water will considerably slow microbial decay.

There is a danger of misidentification. For example, the misidentification of condida spores for starch grains (Haslam 2006b). However, with a large comparative collection and solid experience, this is minimised. My main concern is the misinterpretation of (partly) degraded residues. Deteriorated remains have different properties from those of the original material. Muscle tissue becomes birefringent and starch grains become dull and opaque. Therefore replication studies should be complemented by experimentation, which includes a weathering and deposition

component. Consequently, a residue specialist familiarises himself with degraded materials and he might gain new insights. For example, my work shows that condida spores *are* present in temperate zones (notwithstanding the suggestion of Hardy, Bolus and Conard 2008).

The over- and under interpretation of residues is a complex issue. From this work it is evident that muscle tissue is highly visible on quartz and is probably underrepresented on dark artefacts. Interestingly, because quartz is reflective it is often regarded as more difficult to analyse, but besides muscle tissue, stains are also much clearer on this material than on a dark background. Similarly, white plant tissue is more visible on dark rocks than on light ones. There is no solution to this problem, other than warning specialists that they need to be aware that the colour of the raw materials can bias their results.

Although I argue for large-scale research to contribute to large research questions, I acknowledge that my archaeological samples can only serve as a pilot study. Patterns only become perceptible when large assemblages of similar tools are studied. However, the Sterkfontein sample is a random collection of possibly utilised tools and although extremely unlikely this might have influenced my results. The ancient Sibudu sample, on the other hand, is a selection of artefacts and *pièces esquillées*. Many tools in the Sibudu sample contain muscle tissue and therefore I can say that animal materials were definitely worked at Sibudu. However, I cannot say how this compares to other layers or in what way materials were worked. If I had studied a larger assemblage, perhaps I could have said something about the use of *pièces esquillées*.

The experimental samples contain two tools per individual experiment and, although unlikely, it is possible that two tools are not sufficiently representative of depositional patterns. It would be interesting if other researchers can replicate my results. This would greatly contribute to our insight into residue decay.

8 CONCLUSION

Based on Table 3.12 and Figures 3.2 to 3.9 I made the following predictions:

- The Sterkfontein experiments: organic residues should preserve inside the cave. Outside the cave no organic residues should preserve, but perhaps some resin could.
- The Sterkfontein archaeological sample: no use-related organic residues should preserve.
- The Sibudu experiments: no residues should preserve outside the shelter, but inside organic residues should be present.
- The Sibudu archaeological sample: organic residues should preserve.
- The Wilhelmina Polder experiments: there should be poor organic residue preservation, although plant tissue, starch grains and bone could potentially be preserved.
- Zelhem experiments: no preservation of residues should be possible.

Analysis of the samples in Chapter Four, Five and Six demonstrates that the predictions were correct. Detailed study of taphonomy, curation history and preservation optima lead to correct predictions of residue preservation.

In the Introduction I formulated the following questions about contamination issues:

1. Is there contamination? If so, what is the role of contamination by extraneous residues from the sediment and from post-excavational handling?
2. Do residues move from the sediment onto the stone tools?
3. Can contaminants be distinguished from use-related residues?
4. Is the distribution and association approach by Lombard and Wadley helpful to distinguish use-related residues from contaminants?

Based on research presented in the previous chapters I am able to answer these questions. I found contaminants on all artefacts. Sediment remains can adhere to tools and handling also leaves residues on tools. The moist and used experimental samples showed more sediment contamination than the blanks. It is probable that tools with dry residues show less contamination. Contaminants can be confusing, but they can usually be distinguished. Use-related residues can be differentiated from contaminants

and unintentional residues by quantifying the results and by implementing the contextual approach. The distribution and association approach formulated by Lombard and Wadley (Lombard and Wadley 2007, Wadley and Lombard 2007) is a valuable tool in residue analysis.

I also formulated questions concerning the process of decay:

5. Do muscle tissue, bone, fat, blood and plant starch grains and plant tissue preserve in the same manner?
6. What is the influence of time, precipitation, temperature, burial circumstances, sediment pH, sediment constitution, rock type and processed material on the preservation of residues?
7. Can I make predictions about site selection for analysis from points 5 and 6?
8. Are residues representative of ancient processed materials?

I demonstrated that residue types preserve differently. Plant tissue and bone preserve better than muscle tissue, blood and starch grains. Although fat is a robust material, it is often absorbed into the stone material and sediment and therefore lost for analysis. In addition, what is a good environment for one residue can be destructive for another.

The variables time, precipitation, temperature, burial circumstances, sediment pH, sediment constitution, rock type and processed material influence microbial activity. In general, this means that precipitation leads to decay and that moist environments are bad for preservation. Precipitation also leads to mechanical weathering as residues can be washed off tools. If a soil is rich in nutrients (organic matter) there is more residue decay. At this point I am unable to say if or how the size of sediment grains and aggregates influences residue preservation, but previous studies demonstrated that starch preserves better in sediment with small aggregates. At moderate temperatures (5-35ºC) fewer residues are preserved than at extremely low or high temperatures. However, chemical and mechanical weathering can have a negative effect in extreme temperatures. A moderate sediment pH (pH 6-7.5) leads to decay. Residues are better preserved in extremely acidic or alkaline environments, but chemical and mechanical weathering can have a negative effect on residue preservation in these circumstances. Medium and fine-grained raw materials are better for preservation than large-grained or glass-like materials. At this point it is unclear whether direct burial protects or destroys residues. Direct burial is probably positive for preservation if the sediment shields residues from another harmful environment,

such a humid atmosphere, which is potentially the case at Sibudu, or direct precipitation, as is the case at Wilhemina Polder. Prolonged exposure potentially ensures preservation if it dries the residues out and if it keeps residues safe from microbes. Organic residues either decompose rapidly within a few years or they decompose extremely slowly. Residues always undergo decomposition, but with slow decay it appears that residues can be permanently preserved. Slow decay can take tens of thousands of years, provided that the environments remain stable. In stable environments residues preserve better than in environments that are subjected to reworking and extensive post-depositional processes.

Tools from stable sites with low bioactivity are best for residue analysis. In practice this excludes most open-air sites. Dry and desiccated sites, waterlogged, extremely acidic or alkaline sites and cave sites potentially preserve residues best. On the basis of these results it is possible to make predictions about residue preservation at each archaeological site. The prediction that ancient residues preserve in Sibudu Cave is correct.

Residues can be representative of past processed materials. However, not all residues on tools are use-related and it essential to differentiate between use-related and post-depositional and unintentional remains.

This study has shown that residue-analysis must be conducted and interpreted with caution. Unfortunately the taphonomy of many sites precludes the preservation of ancient residues. I have, however, shown that it is possible to make predictions about which sites are suitable for residue analysis and this enables the analyst to save time selecting sites and ancient samples. When sites and artefacts are carefully selected based on feasibility studies, I argue that residue-analysis can be a valuable tool for the reconstruction of processed materials and past activities.

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SUMMARY

I investigate the decay mechanisms for microscopic organic residues. Residue analysis aims to identify microscopic remains or traces that are left on a tool's surface after use. Analysts have identified organic remains such as plant starch grains, fibres, tissue cells, resin; animal blood films and red blood cells, muscle tissue, connective fibres, bone fragments, hairs; fish scales, feathers and shell. Besides organic residues, it is also possible to distinguish inorganic deposits such as ochre and ash. After the processed materials are identified on the artefact, residue analysis can be used, for example, to reconstruct tool use and assess site function.

Preservation of residues on artefacts is a debated topic and I decided to investigate the following issues:

- Non-use-related residues, for example from unintentional handling or sediments, might contaminate artefacts.
- Researchers are unable to describe the mechanism of residue preservation.

My study investigates what research biases might occur from extraneous, or "extra", remains and from deteriorated, or "missing" residues.

I use experimental archaeology and middle-range theory (Binford 1983, Raab and Goodyear 1984, Schiffer 1988, Reynolds 1999, Outram 2008) to bridge the gap between the apparently static archaeological data (what one sees on the tools) to a dynamic reconstruction of past processed materials (what tools were used on and for). The aim of this study is to produce predictions about residue preservation, recognition and possible misinterpretation.

In order to understand the decay of organic residues, I set out experimentally made tools with modern residues at Sterkfontein Caves and Sibudu Cave in South Africa and at Wilhelmina Polder and Zelhem in the Netherlands. The modern flakes were of chert, hornfels and flint respectively and they were used to process muscle tissue, bone, plant tissue, starch rich and plant material. The samples at the sites were subdivided and deposited under different variables; these included time, precipitation, temperature, burial circumstances, sediment pH, sediment constitution, rock type and processed material. Practically this means that part of the sample was deposited inside the caves and the other part outside and that part of the sample was deposited on top

of the soil and another part was buried. The sediment pH at Zelhem is low, whereas the pH at Wilhelmina Polder is high; at Sterkfontein and Sibudu the sediments are neutral; the sediment at Sterkfontein consists of sandy clay, the sediment at Sibudu consists of anthropogenic ash, at Wilhelmina Polder the sediment is clayey and at Zelhem it is sandy. After four weeks I excavated and analysed some of these tools from Sterkfontein and Sibudu. After one year I analysed the remaining set. The Zelhem and Wilhelmima Polder experiments were all collected and analysed after six months. At Zelhem and Wilhelmina Polder the experiments also included sets of sandpaper of different grain sizes and glass slides with modern residues. Part of this sample was buried with the residues facing up and the other part was facing down. This was to test to influence of the raw material surface and microenvironment on residue preservation. All experiments had a set of blank doubles; these are unused flakes, which were deposited in the same settings. These blanks served as control sample and they helped me to study the distribution of contaminants on stone surfaces.

I also analysed ancient samples of stone tools from Sterkfontein and Sibudu. This was to study the possible correlation in preservation between the experimental and ancient residues.

I formulated predictions about the preservation of residues under different circumstances, and in the course of this study I demonstrated that reliable predictions can be made when the taphonomy and curation history of tools and preservation optima of organic remains are carefully studied.

This study clearly demonstrates that all experimental and ancient artefacts contain contaminants. Sediment remains adhere to tools and handling also leaves residues on tools. Not only plant type residues are possible contaminants, I also found contaminant animal type remains, such as connective fibres and muscle tissue. The used samples showed more sediment contamination than the blanks. Because the moist, use-related residues act as an adhesive it is probable that tools with dry residues show less contamination.

It is possible to distinguish use-related residues from contaminants and unintentional residues by quantifying the results and by implementing the contextual approach. Use-related residues are more abundant than contaminants. In addition, use-related residues form coherent distribution patterns with suites of associated residues. For example, processing of animal material leaves deposits of muscle tissue and fat

around the working edge. The distribution and association approach formulated by Lombard and Wadley (Lombard and Wadley 2007, Wadley and Lombard 2007) is a valuable tool in residue analysis.

Different residue types preserve differently. Plant tissue and bone preserve better than muscle tissue, blood and starch grains. Although fat is a tough material, it is often absorbed into the stone material and sediment and therefore lost for analysis. In addition, what is a good environment for one residue can be destructive for another. For example, at Sterkfontein (outside the cave), almost no starch residues were preserved on the starch tools, but at Wilhelmina Polder plenty of starch grains were present and, in addition, many use-related epidermal tissue deposits were also preserved. Another example is muscle tissue, which preserved well inside Sibudu Cave, but preserved badly outside the cave. Outside Sibudu Cave, bone and plant tissue deteriorated less than muscle tissue.

Microbial activity is the main reason for deterioration. From the experiments I gained insight into how the variables time, precipitation, temperature, burial circumstances, sediment pH, sediment constitution, rock type and processed material affect microbial activity. Consequently, precipitation leads to decay and therefore moist, humid and fluctuating environments are bad for preservation. Precipitation also leads to mechanical weathering as residues can be washed off tools. When a soil is rich in nutrients (organic matter) there are more microbes in the sediment and as a result more residue decay. At moderate temperatures less residues are preserved than at low (<5ºC) or high temperatures (>35ºC). However, chemical and mechanical weathering can have a negative effect in more extreme temperatures. Because microbes flourish in environments with a moderate pH (pH 6-7.5), these settings lead to more decay. Residues are better preserved in acidic or alkaline environments, but chemical and mechanical weathering sometimes benefit from more extreme pH values and can then have a negative effect on residue preservation. Medium- and fine-grained rocks are better for preservation than large-grained or glass-like materials. At this point it is unclear if direct burial protects or destroys residues. Direct burial probably promotes preservation if the sediment shields residues from another harmful environment, such a humid atmosphere (potentially the case at Sibudu), or precipitation (as is the case at Wilhemina Polder). Prolonged surface exposure may ensure preservation if it dehydrates the residues and if it keeps residues safe from microbes. Residues always undergo decomposition. Depending on the circumstances,

organic residues either decompose rapidly within a few years or extremely slowly. With rapid decay, no residues are left for analysis even after only one year. With slow decay it appears that residues are permanently preserved. However, chemically and sometimes visually, these residues undergo change. Slow decay can take up to tens of thousands of years, provided that the environments remain stable. In a stable environment, residues preserve better than in an environment that is subjected to reworking and extensive post-depositional processes.

The described results imply the following procedures for residue analysis: tools should be selected from stable sites with low bioactivity. In practice this excludes most open-air sites. Dry and desiccated sites, waterlogged, extremely acidic or alkaline sites and cave sites potentially preserve residues best. On the basis of these results it is possible to make predictions about residue preservation for each site. For example, the prediction made in this study that ancient residues are preserved inside Sibudu Cave proved correct.

Selected artefacts must be handled with care and residue analysis should be the first analysis to be conducted on a tool sample. This means that no lithic or other analysis should take place prior to residue analysis. Sediment samples and blank flakes should be analysed to gain insight into possible contaminants and the raw material.

The selected sample should include a substantial number of artefacts. Because residual decay always occurs, some tools in a sample will not show the expected patterning or preserve residues. To filter out these exceptions, and to ensure that the overall signal becomes clear, more than 30 tools should be analysed. In addition, these should be tools of the same type. It is only possible to recognise and compare distribution patterns when dealing with a uniform sample. For example, possible hafting of scrapers and the accompanying distribution pattern and residue suite is easier to recognise in a scraper-only-sample than in a mixed sample including retouched flakes and points.

The contextual approach should be applied and residues should be quantified. The deductive list, which is discussed in detail in Chapter Six and Seven, is a practical and heuristic device for conducting residue analysis and it follows the contextual approach.

Residues can be representative of past processed materials. However, not all residues on tools are use-related and it essential to differentiate between use-related and post-depositional and unintentional remains.

This study has shown that residue-analysis must be approached with caution. Unfortunately the taphonomy of many sites precludes the preservation of ancient residues. However, with the aid of the predictions regarding circumstances of preservation of residues, the careful selection of sites and samples, and by implementing the contextual approach, I have argued that residue-analysis can be a valuable tool in the reconstruction of processed materials and past activities.

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APPENDICES

Appendix A: Chapters Two and Three

In this appendix the figures belonging to Chapters Two and Three are presented. Appendix A includes photographs of replicated and experimental residues, sediment samples and the rock types. Appendix A is available on the CD-ROM at the back of this thesis.

Appendix B: results from the experimental sample

In Appendix B the micrographs and distribution maps from Chapter Four are presented. This appendix covers the visual data from the experiments. Appendix B is available on the CD-ROM at the back of this thesis.

Appendix C: Results from the ancient Sterkfontein sample

In this appendix the figures belonging to Chapter Five are presented. Appendix C covers the results from the ancient Sterkfontein sample and includes micrographs of the found remains and distribution maps. Appendix C is available on the CD-ROM at the back of this thesis.

Appendix D: Results from the ancient Sibudu sample

The figures in Appendix D belong to Chapter Six. The micrographs of residues and the distribution maps cover the results from the ancient Sibudu sample. Appendix D is available on the CD-ROM at the back of this thesis.

SUPPLEMENT: CD-ROM WITH APPENDICES AND ELECTRONIC COPY OF THESIS

Appendices A-D are stored on the provided CD-ROM. In addition, a copy of this thesis is also available in PDF format on the disk.

A APPENDIX: CHAPTERS TWO AND THREE

This appendix deals with the micrographs from Chapter Two and Three. Unless stated otherwise, the pictures are taken from experimental and replicated modern tools with residues or sediment samples. All pictures are taken in darkfield light using no filters, unless stated otherwise.

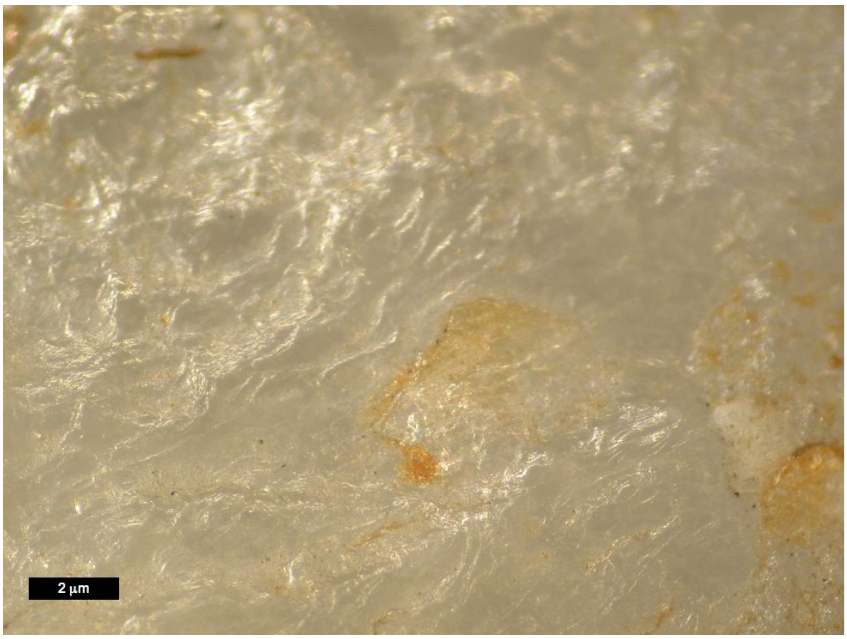


Figure A.1: Micrograph of translucent quartz surface with some red sediment smears. Magnification 50.



Figure A.2: Micrograph of cortex surface on hornfels artefact. Magnification 50.



Figure A.3: Micrograph of fatty flint surface. Magnification 50.

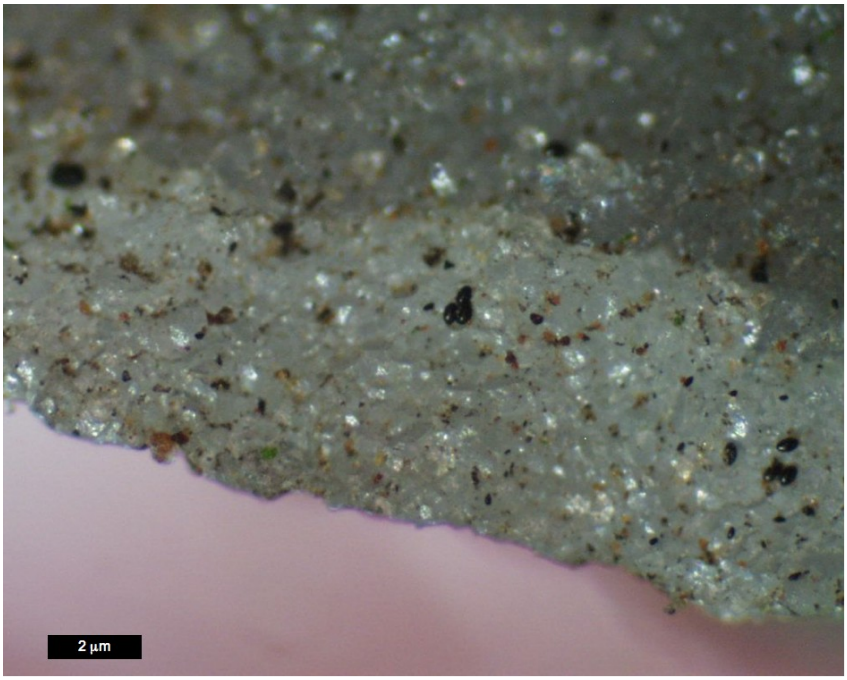


Figure A.4: Micrograph of chert surface. Magnification 50.

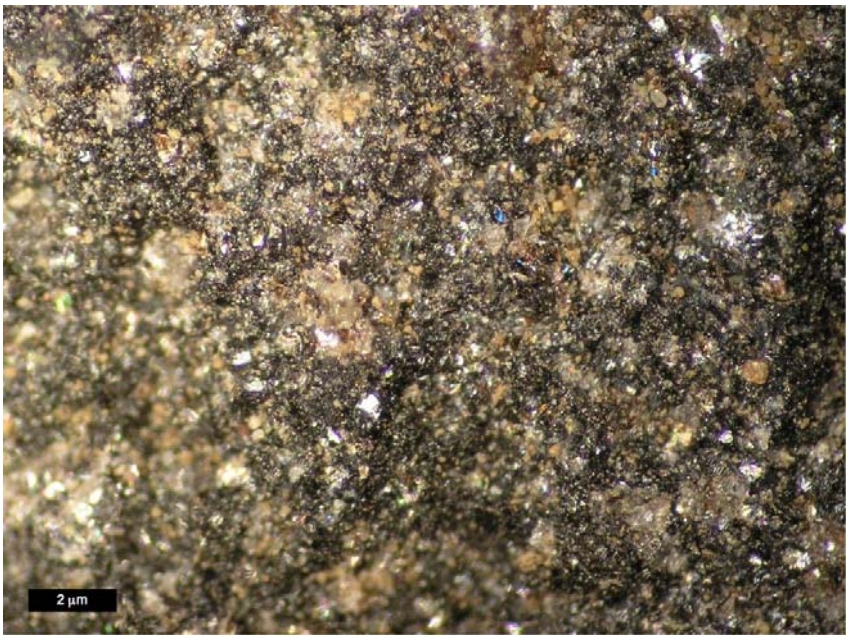


Figure A.5: Micrograph of hornfels surface. Magnification 50.

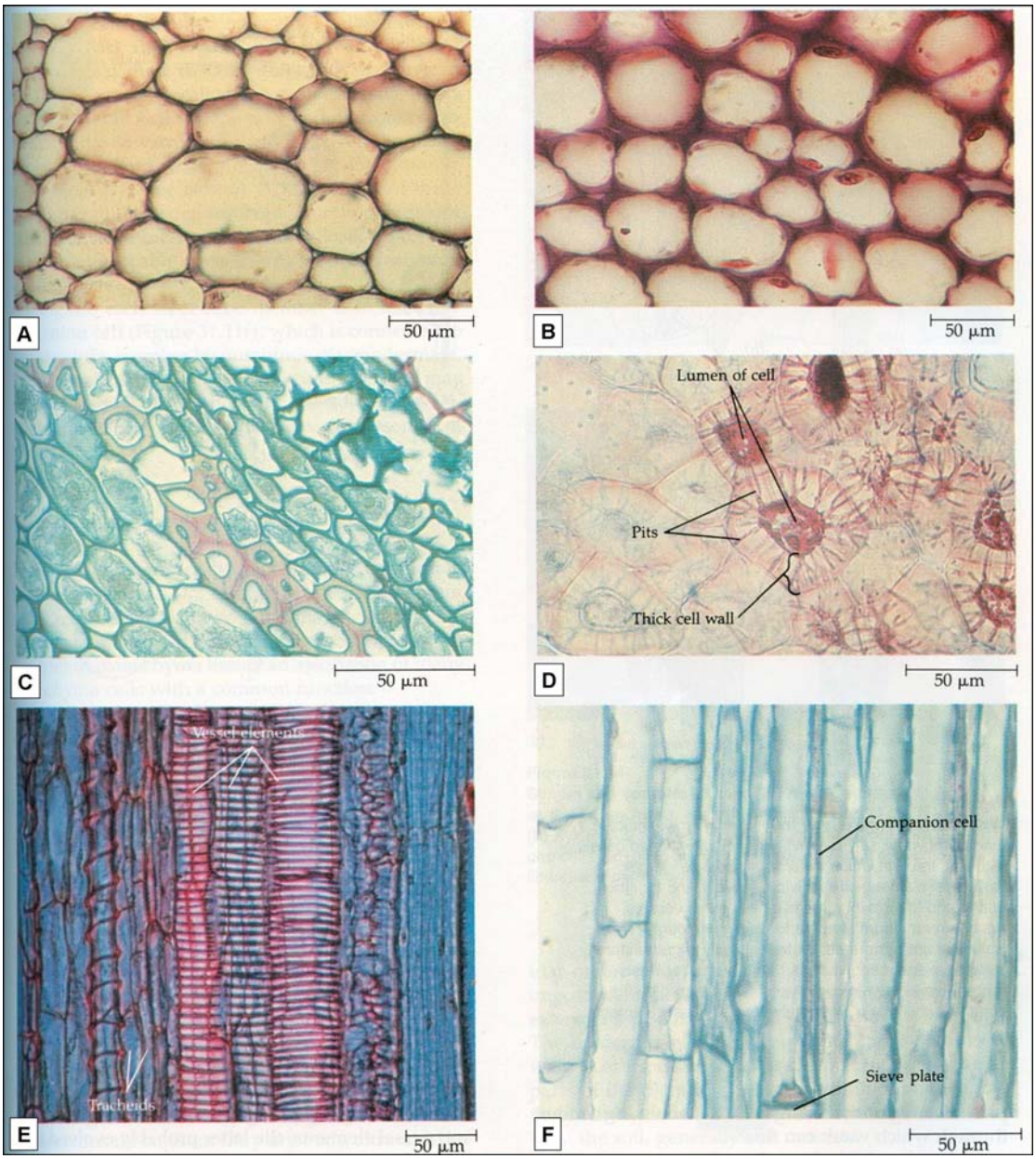


Figure A.6: Types of plant cells (E and F are in longitudinal section and the other pictures are in cross section). A= parenchyma cells; B= collenchyma cells; C= sclerenchyma cells; D= slereid cells; E= waterconducting cells of xylem; F= nutrient conducting cells of phloem. Adapted from Campbell 1990, Figure 31.11, page 689.

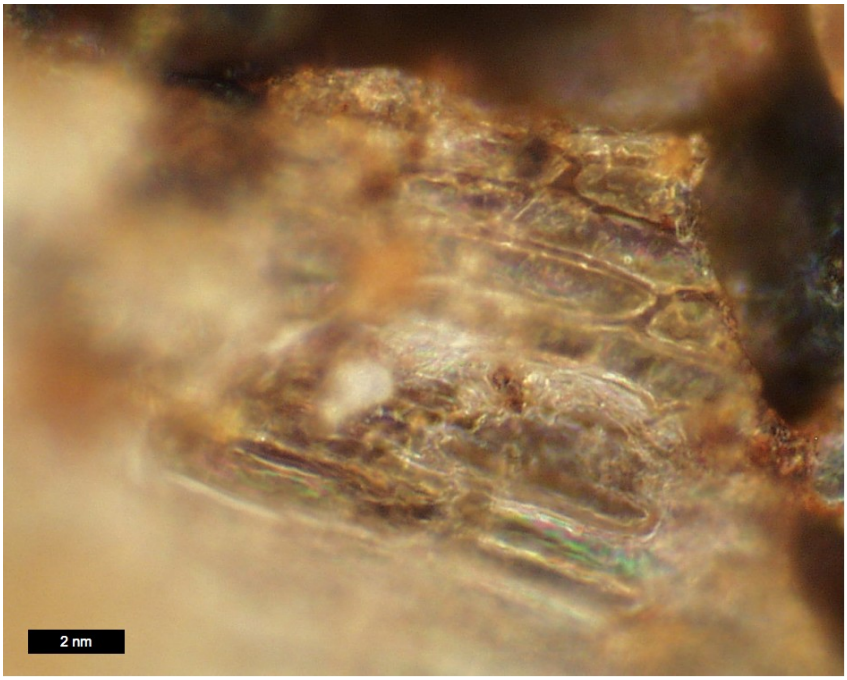


Figure A.7: Micrograph of elongated plant cells. Note the cell walls. Magnification 500, brightfield with polarised light.

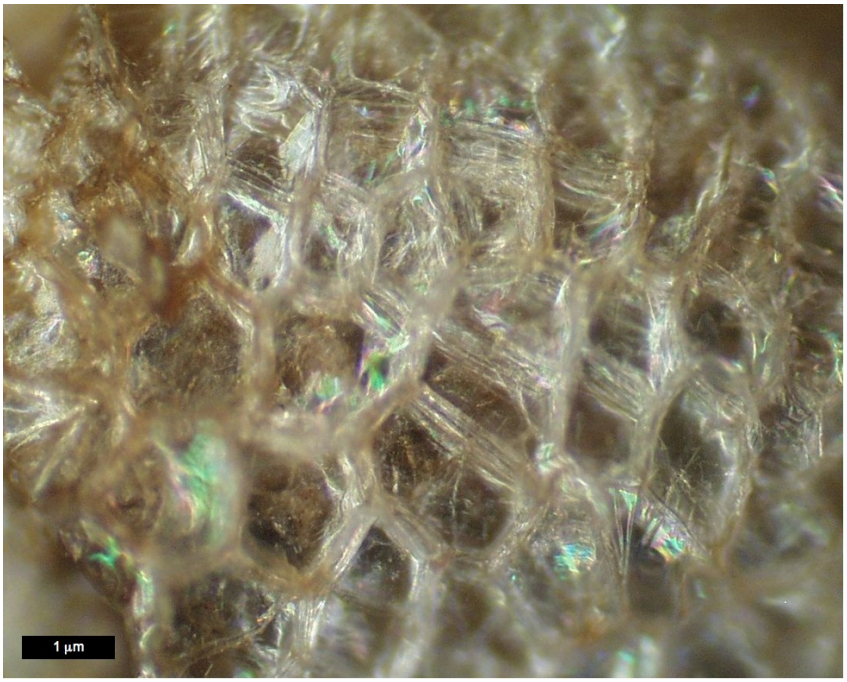


Figure A.8: Micrograph epidermal tissue. Note the thick cell walls and the rigid organisation of the tissue. Magnification 100.



Figure A.9: Micrograph of plant fibres. Note that these fibres are more rigid and translucent than the muscle fibres in A.54 and A.55. Magnification 100.

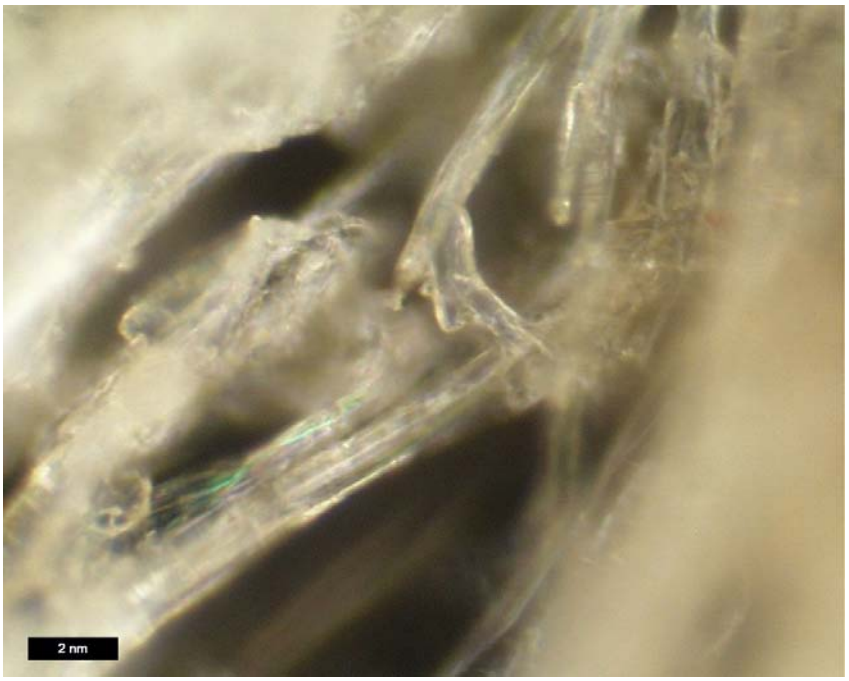


Figure A.10: Micrograph plant fibres. Note that the fibres have a translucent quality and are birefringent. Magnification 500.



Figure A.11: Micrograph of a modern blue fibre. Magnification 50.

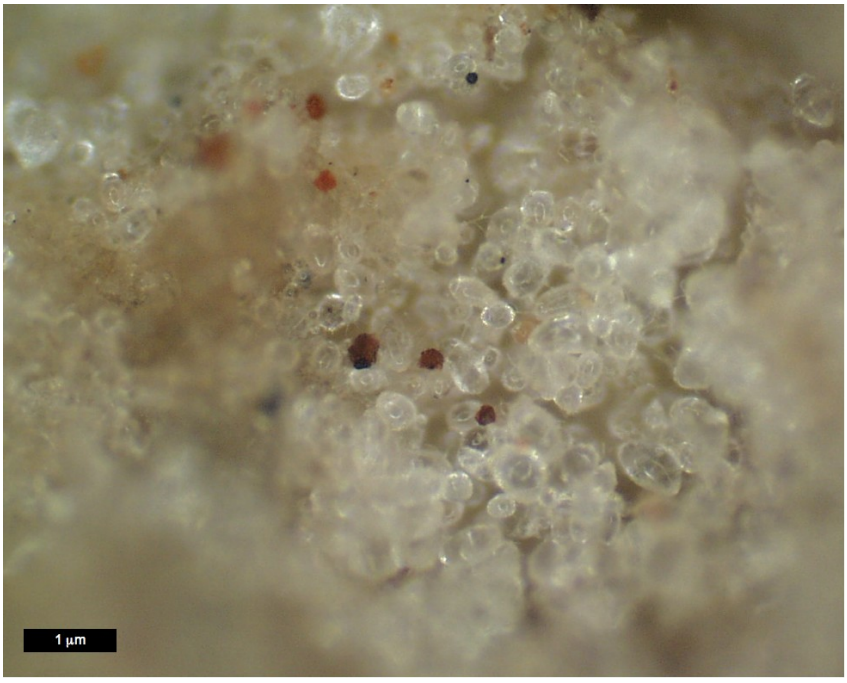


Figure A.12: Micrograph of starch grains. Magnification 100.

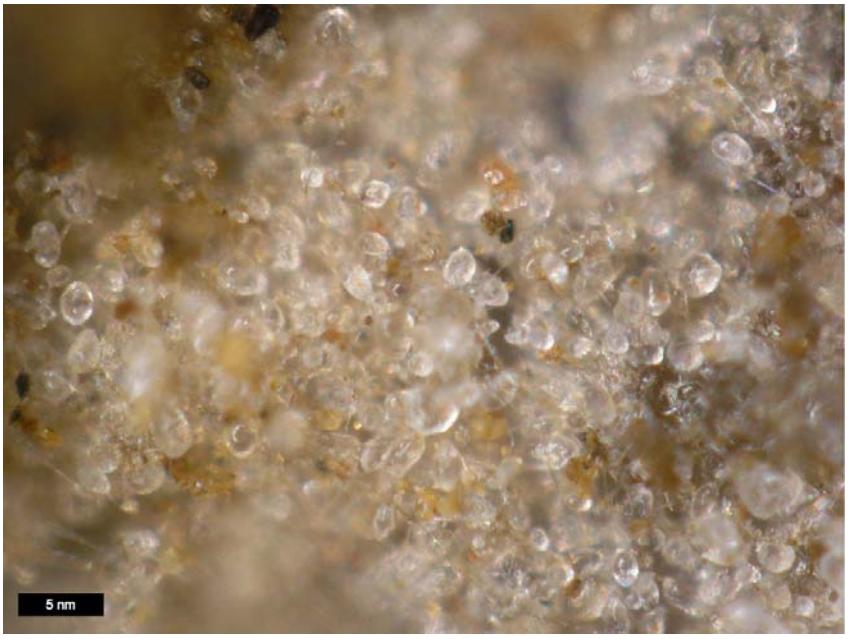


Figure A.13: Micrograph of slightly birefringent starch grains. Magnification 200.

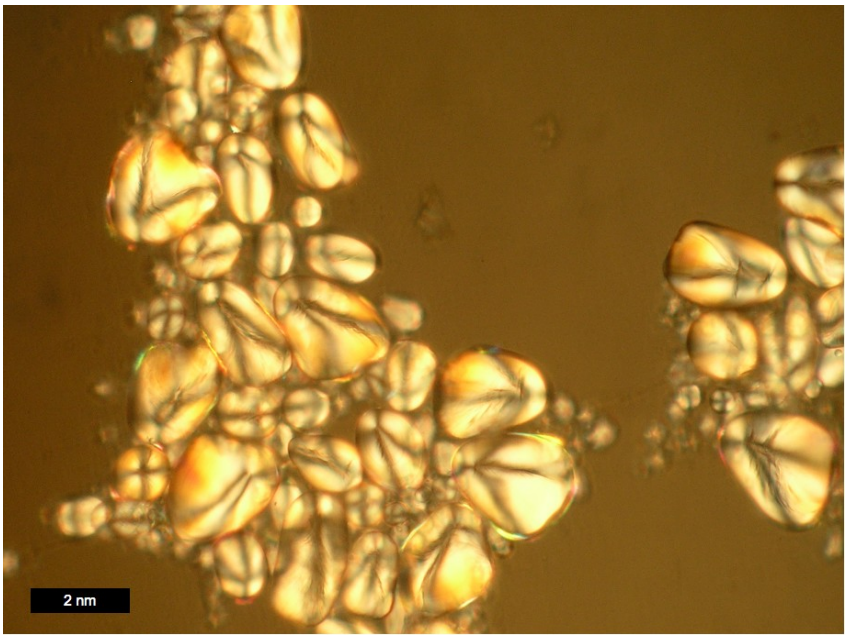


Figure A.14: Micrograph of potato starch grains. Note the characteristic extinction cross. Magnification 500, brightfield polarised light.



Figure A.15: Micrograph of white degraded plant tissue. Note that this tissue has a blue gloss, similar to fat. Also note the angular and elongated organisation are remnants from the structured cells. Magnification 50.

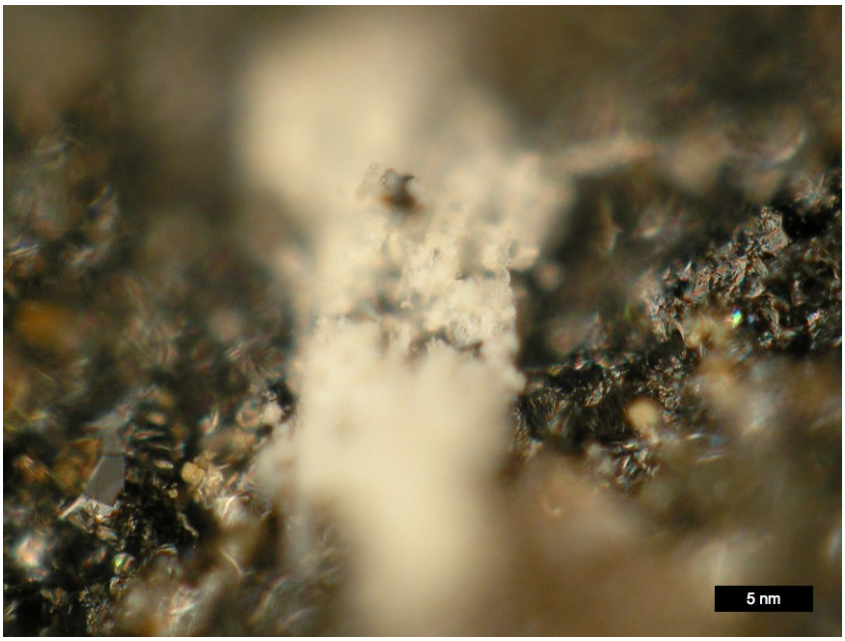


Figure A.16: Micrograph of white degraded plant tissue. Note the angular and elongated organisation are remnants from the structured cells. Magnification 200.

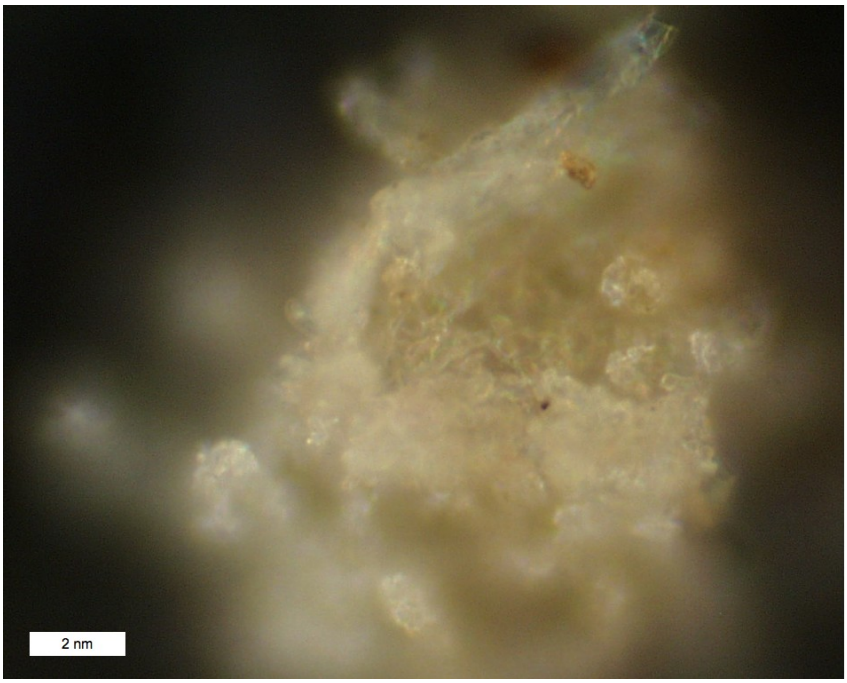


Figure A.17: Micrograph of white degraded plant tissue. Note that the degraded tissue has a bone and fatty appearance too. Magnification 500.

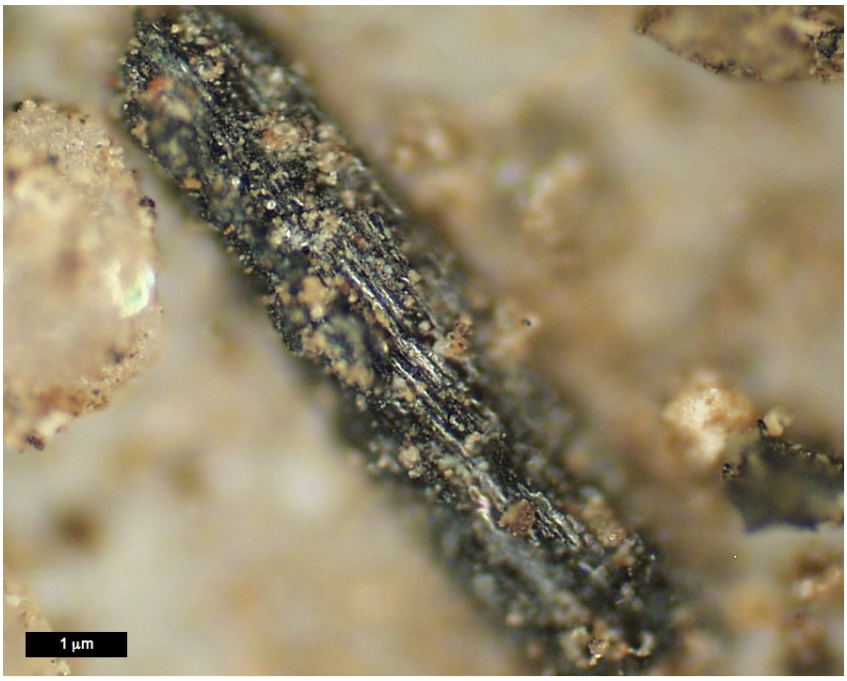


Figure A.18: Micrograph of black degraded plant tissue (charcoal). The plant cells and the rigid organisation of the tissue are still visible. Magnification 100.



Figure A.19: Micrograph of black degraded epidermal plant tissue. The plant cells and the rigid organisation of the tissue are still visible. Note that the tissue is birefringent. Magnification 200.



A



B

Figure A.20: (A) Pristine resin bleeding from a tree, photo by J. Rursch (2006), Wikimedia Commons. (B) Karoo acacia gum (*Acacia karroo*) (left) and ground resin (right) (courtesy of Lyn Wadley).



Figure A.21: Acacia (*Acacia* sp.) resin/gum. Note the cracked surface. Magnification 50 (courtesy of Lyn Wadley).

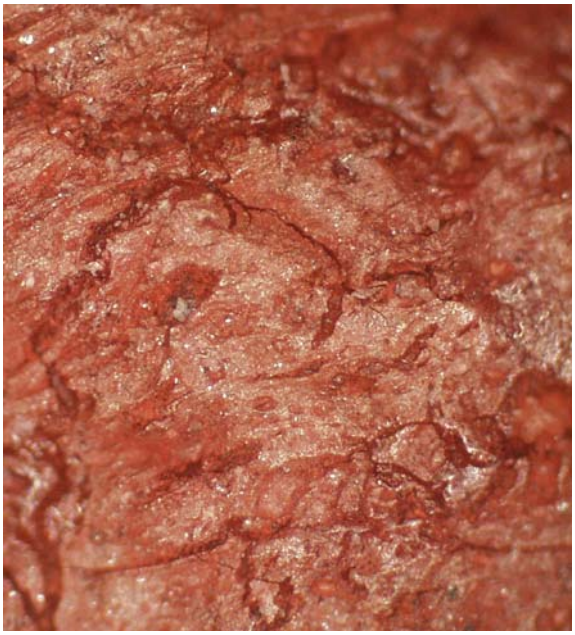


Figure A.22: A heated mixture of acacia resin/gum and ochre. This resin mixture is more organic and flowing than pristine resin. Magnification 50 (courtesy of Lyn Wadley).

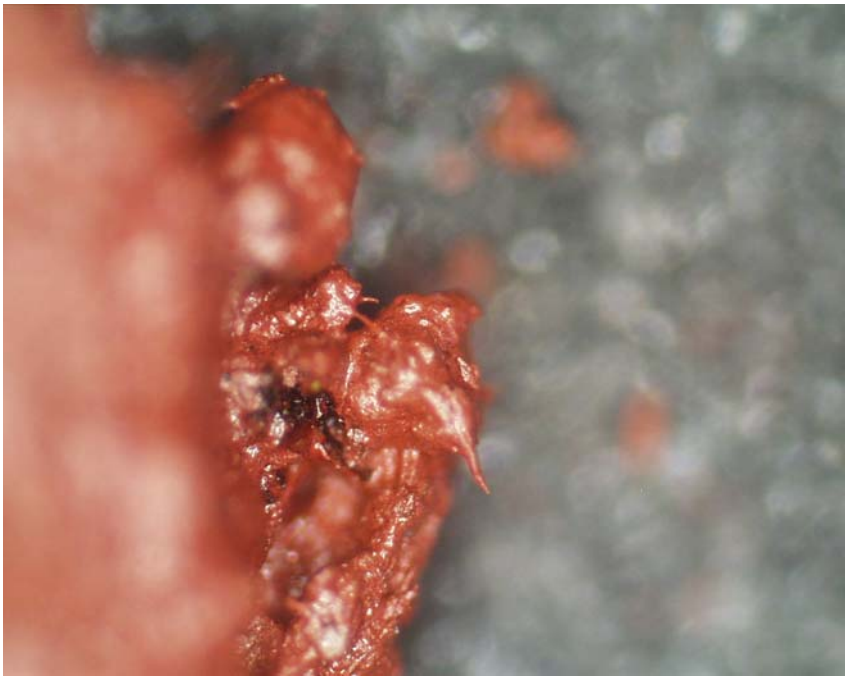


Figure A.23: Micrograph a mixture of ochre and acacia gum/resin. This mixture is more organic and has a sticky feel and is not glass-like anymore like pristine resin. Magnification 50 (courtesy of Lyn Wadley).



Figure A.24: Micrograph of plant exudate from courgette (*Cucurbita pepo)*. Photo by F. Vincentz (2007), Wikimedia Commons.

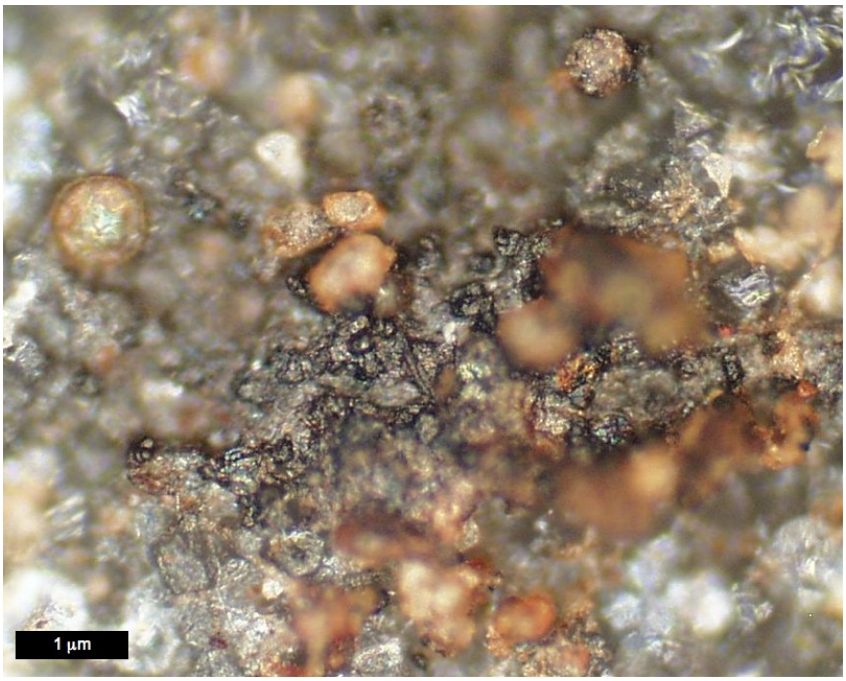


Figure A.25: Micrograph of a plant exudate on a stone surface. Magnification 100.



Figure A.26: Micrograph of a rootlet. Magnification 50.



Figure A.27: Micrograph of a deteriorated rootlet. Magnification 50.

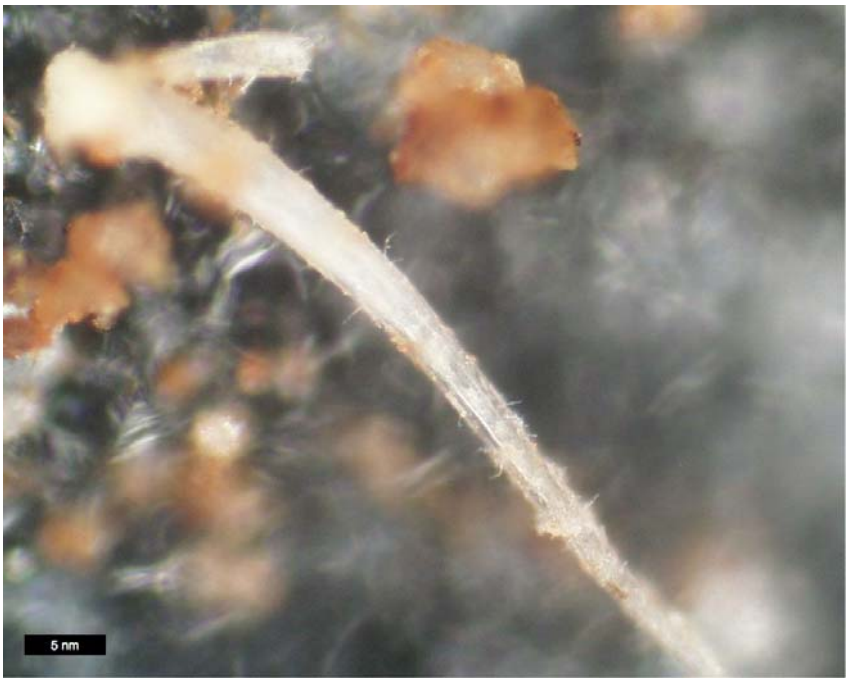


Figure A.28: Micrograph of a rootlet. Note the hair-like roots that grow from the main branch; these are indicative for root parts and are not present on plant fibres. Magnification 200.

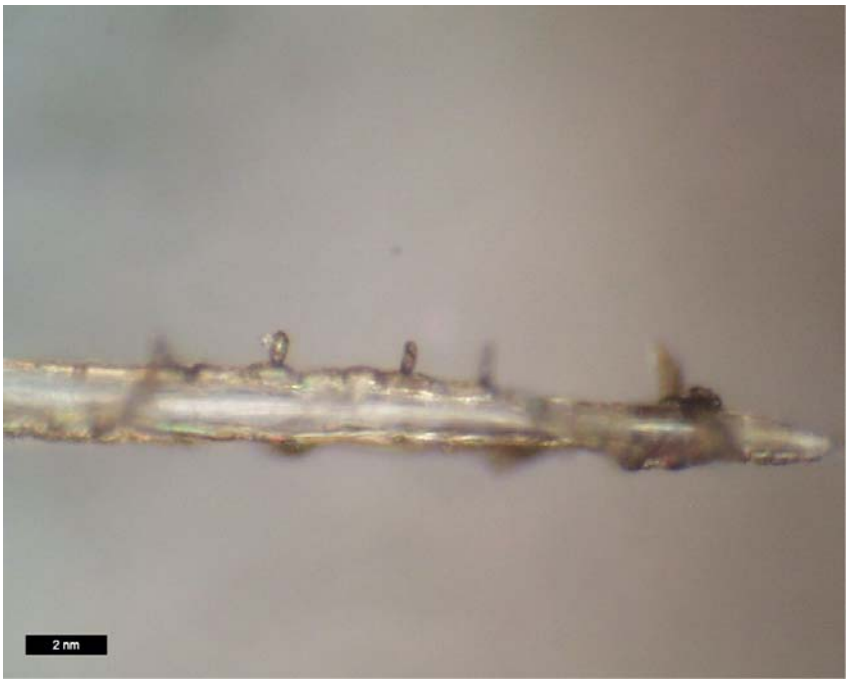


Figure A.29: Micrograph of a rootlet. Note the hair-like roots growing from the main root. Magnification 500.

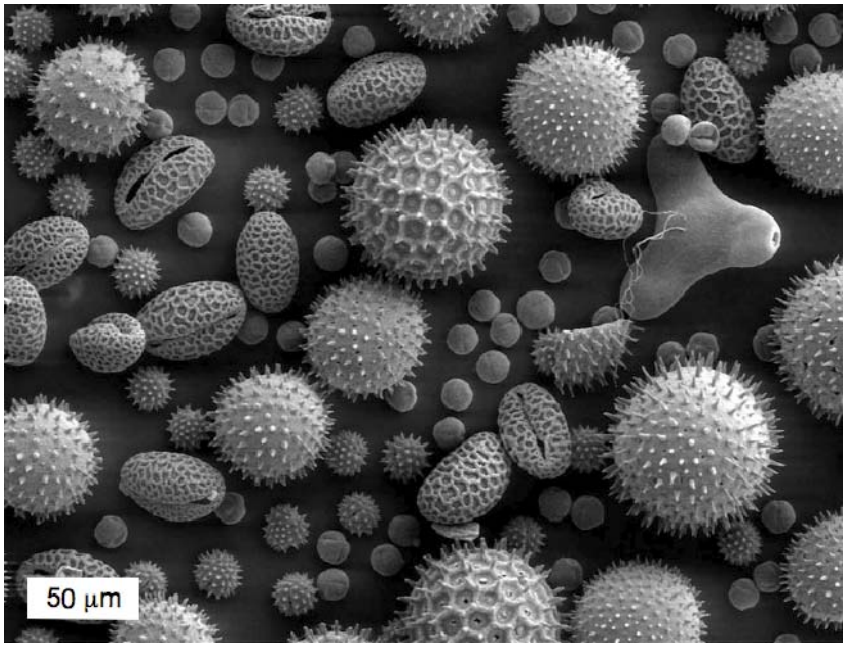


Figure A.30: SEM image of pollen from sunflower (*Helicantuus annuus*), morning glory (*Ipomoea purpurea*), hollyhock (*Sildalcea malviflora*), lily (*Lilium auratum*), primrose (*Oenothera fruticosa*) and castor bean (*Ricinus communis*). Magnification 500. Image by Dartmouth Electron Microscope Facility, Dartmouth College (2004), Wikimedia Commons.

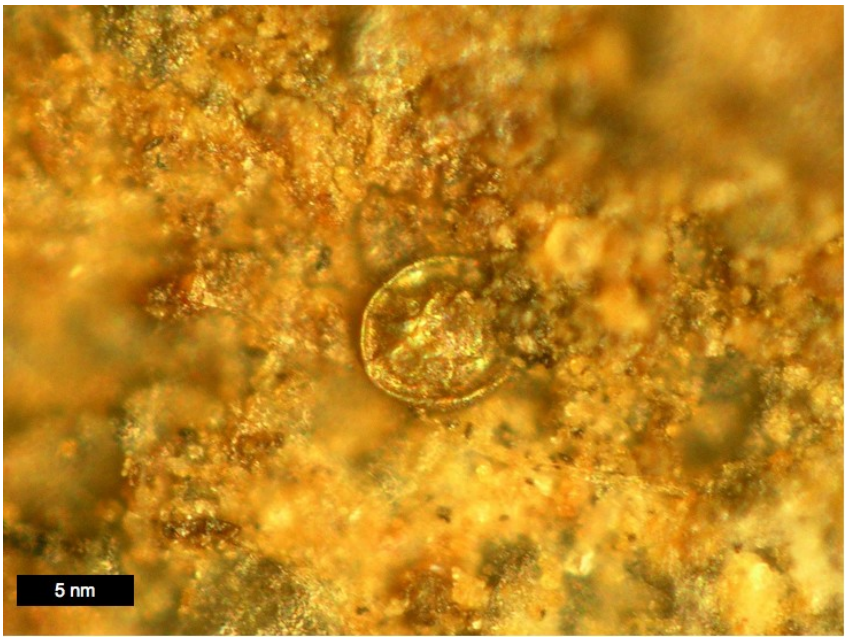


Figure A.31: Micrograph of a pollen grain. Magnification 200.

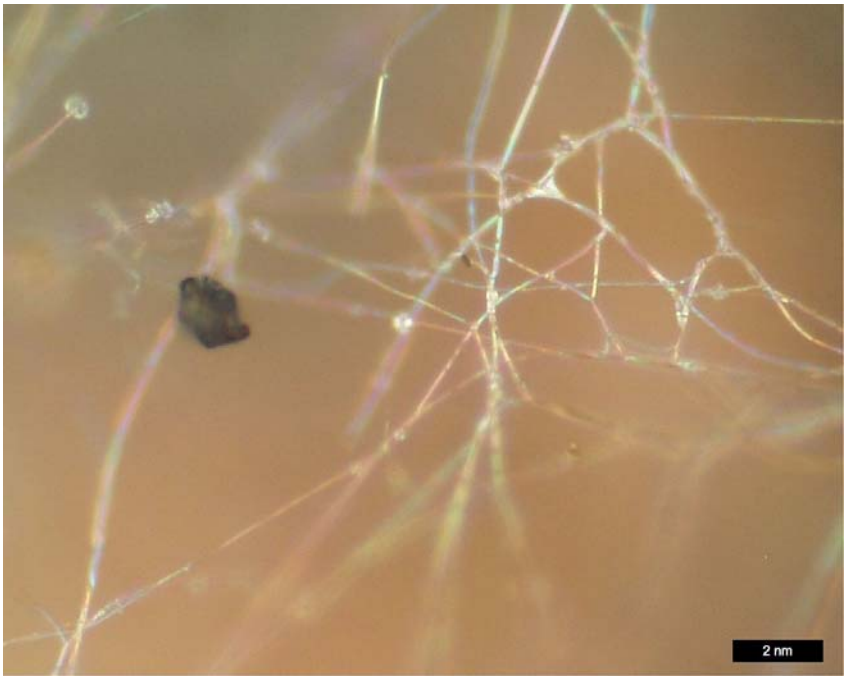


Figure A.32: Micrograph of birefringent hyphae. Magnification 500, brightfield cross-polarised light.

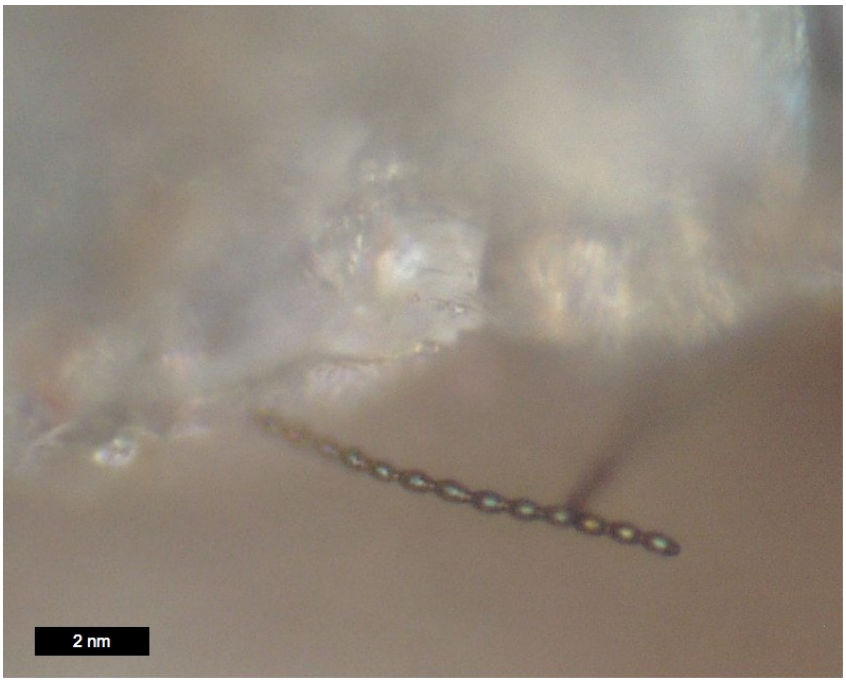


Figure A.33: Micrograph of a fungus fruiting body. Magnification 500.

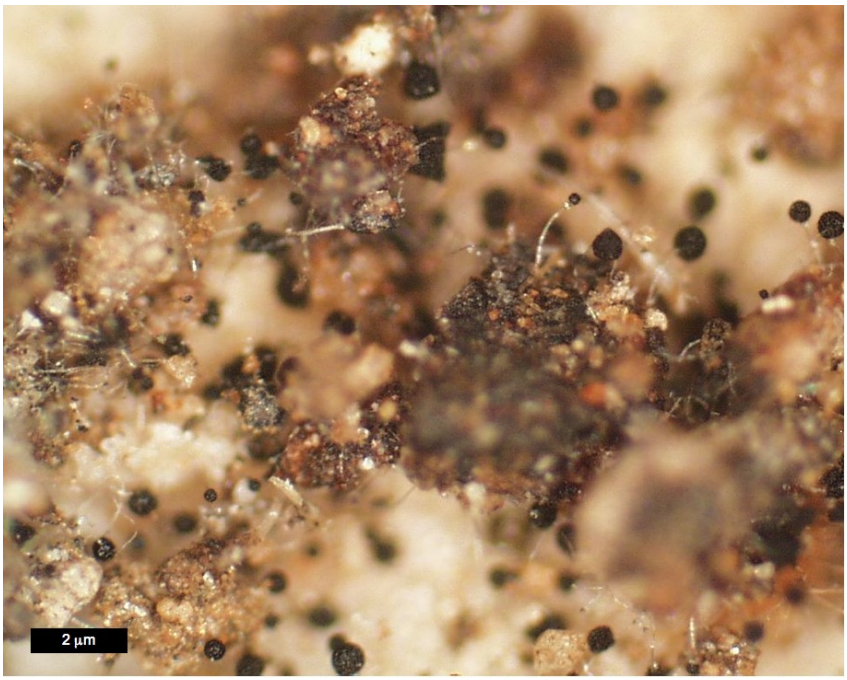


Figure A.34: Micrograph fungus fruiting bodies. Magnification 50.

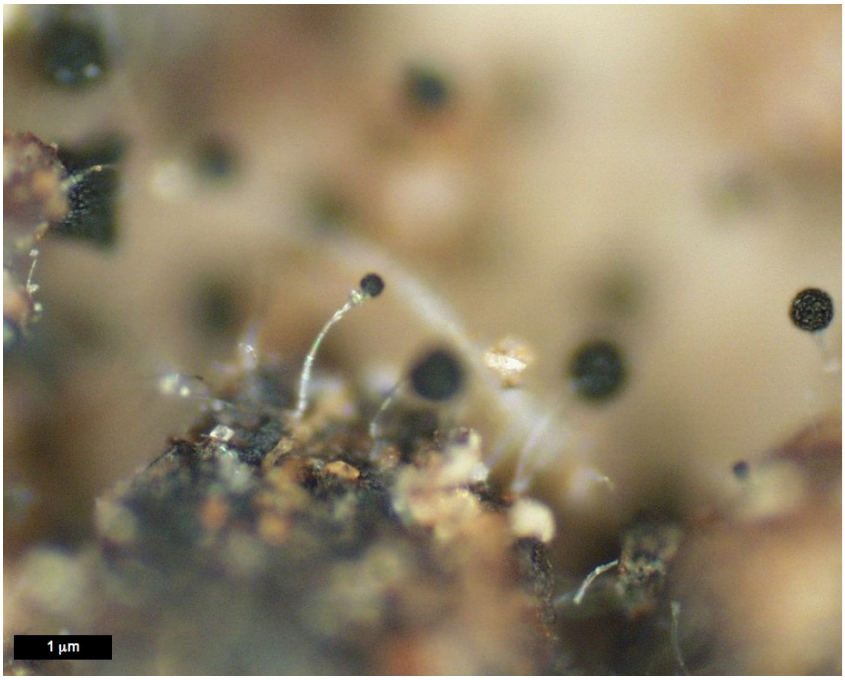


Figure A.35: Micrograph of fungus fruiting bodies. Magnification 100.



Figure A.36: Micrograph of a fungus mycelium. Magnification 200.

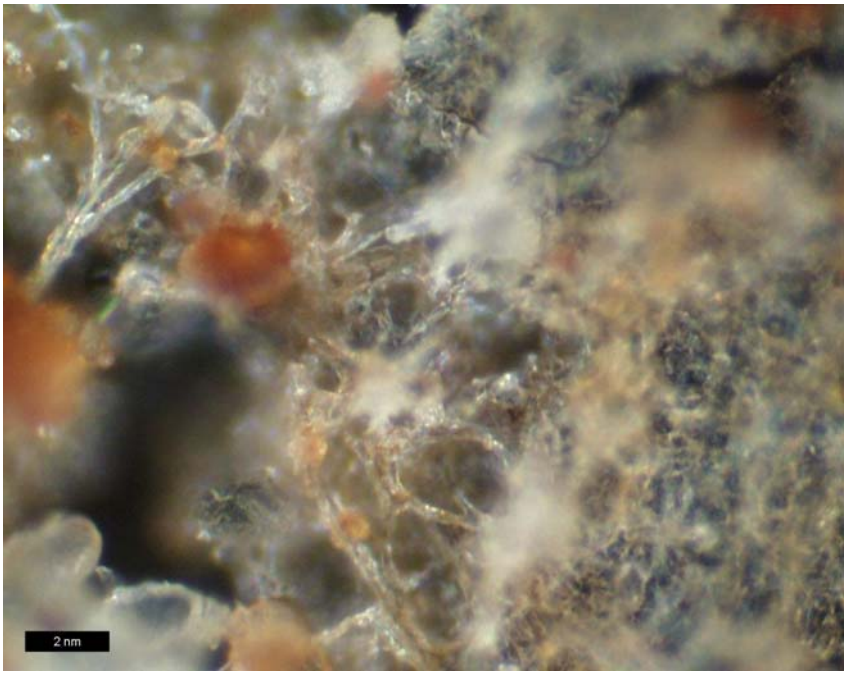


Figure A.37: Micrograph of "gelatinous" hyphae. This hypha resembles "gelatinous" muscle tissue as in A.52. Magnification 500.

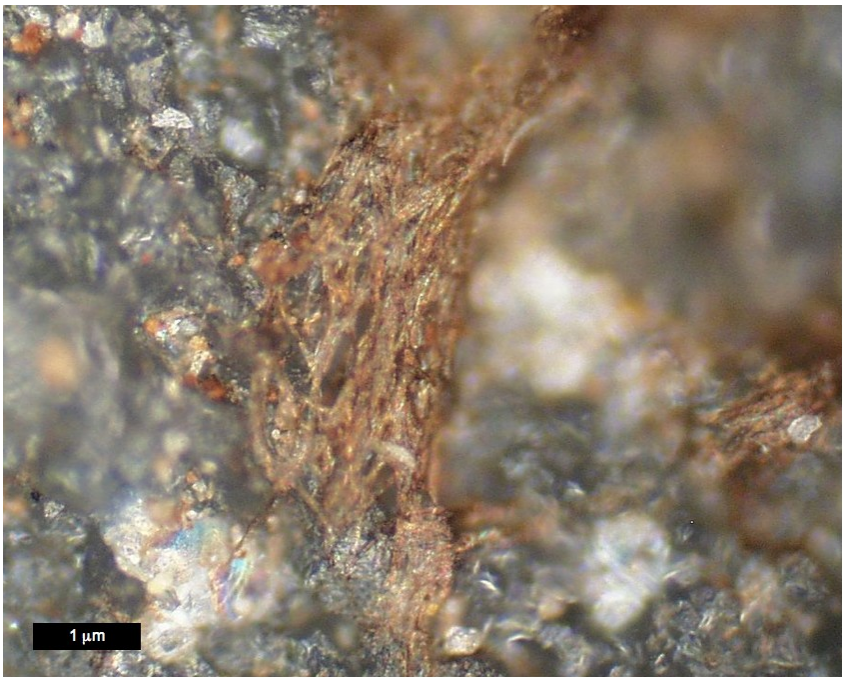


Figure A.38: Micrograph of brown fungus mycelium. Magnification 100.

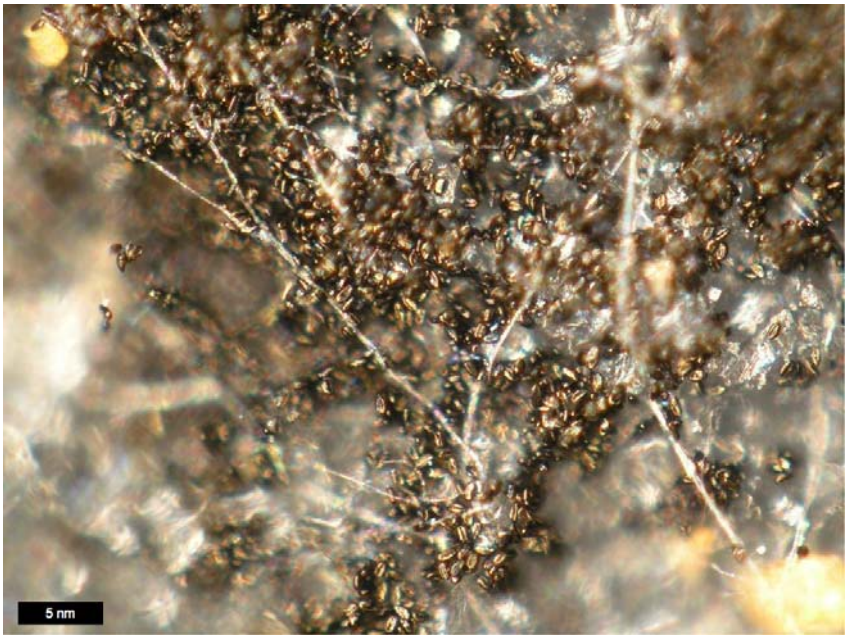


Figure A.39: Micrograph of black spores. Magnification 200.

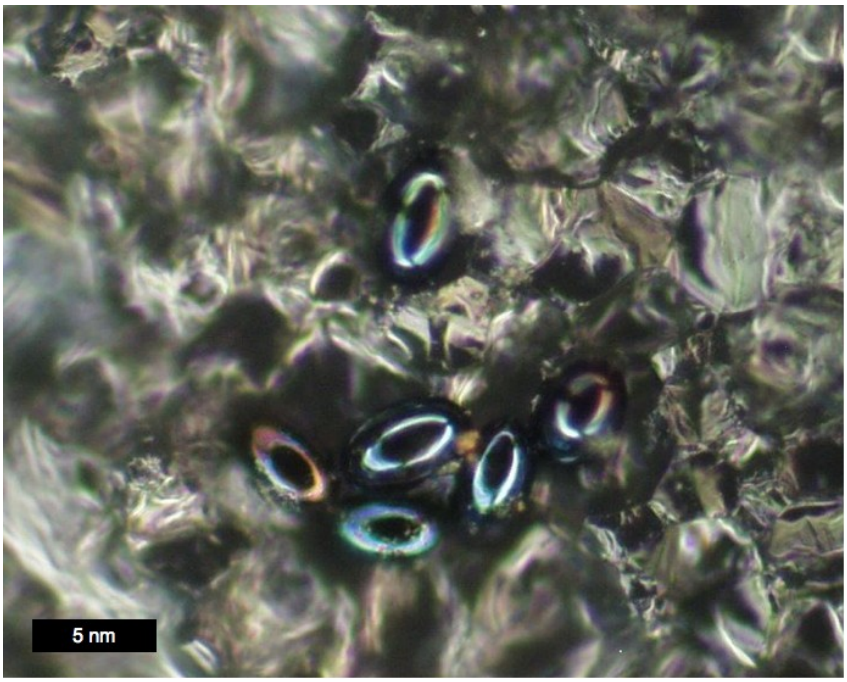


Figure A.40: Micrograph of black spores. Magnification 200.

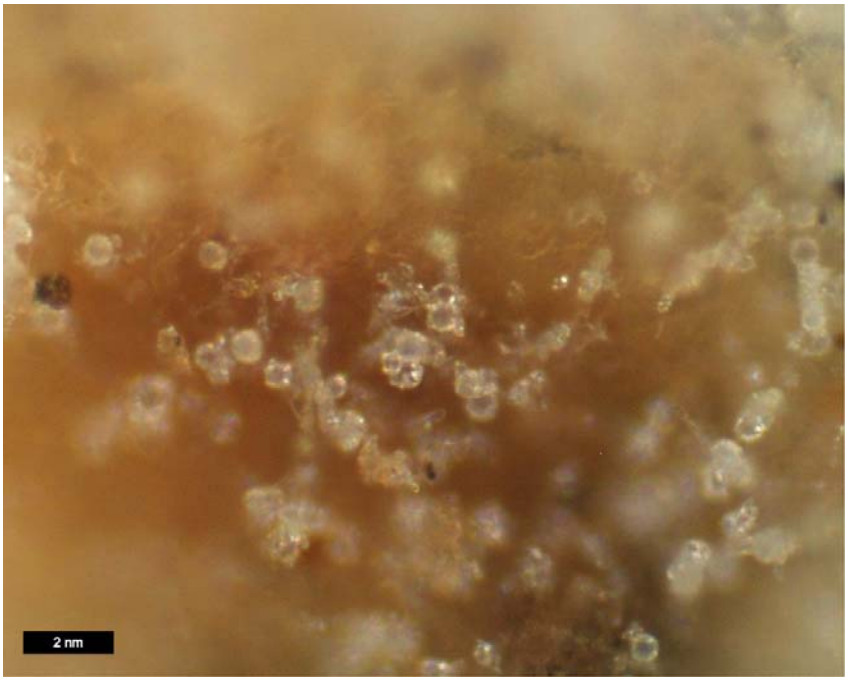


Figure A.41: Micrograph of white spores. This type of spores can easily be mistaken for starch grains. Magnification 500, brightfield polarised light.

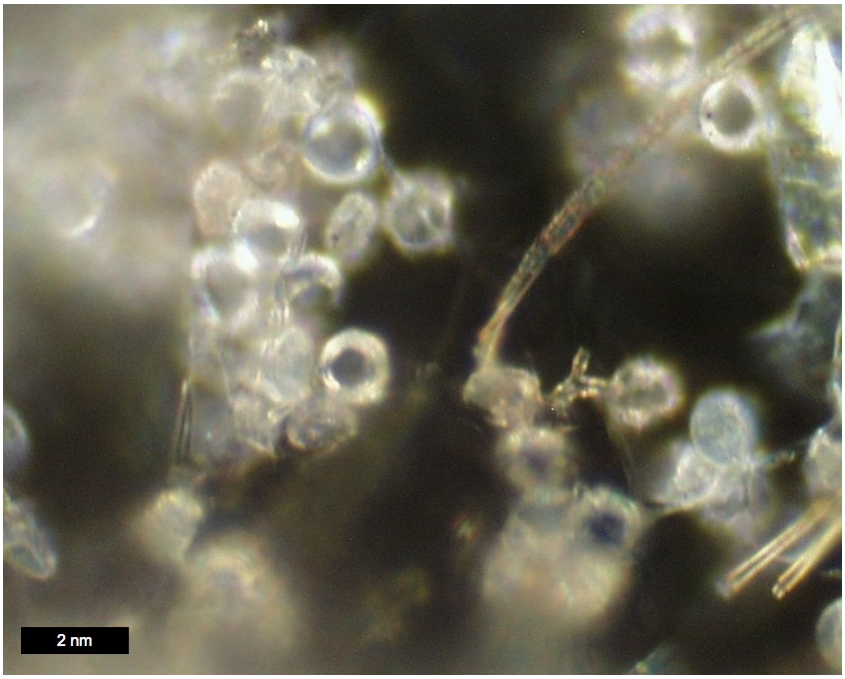


Figure A.42: Micrograph of white spores. Magnification 500.

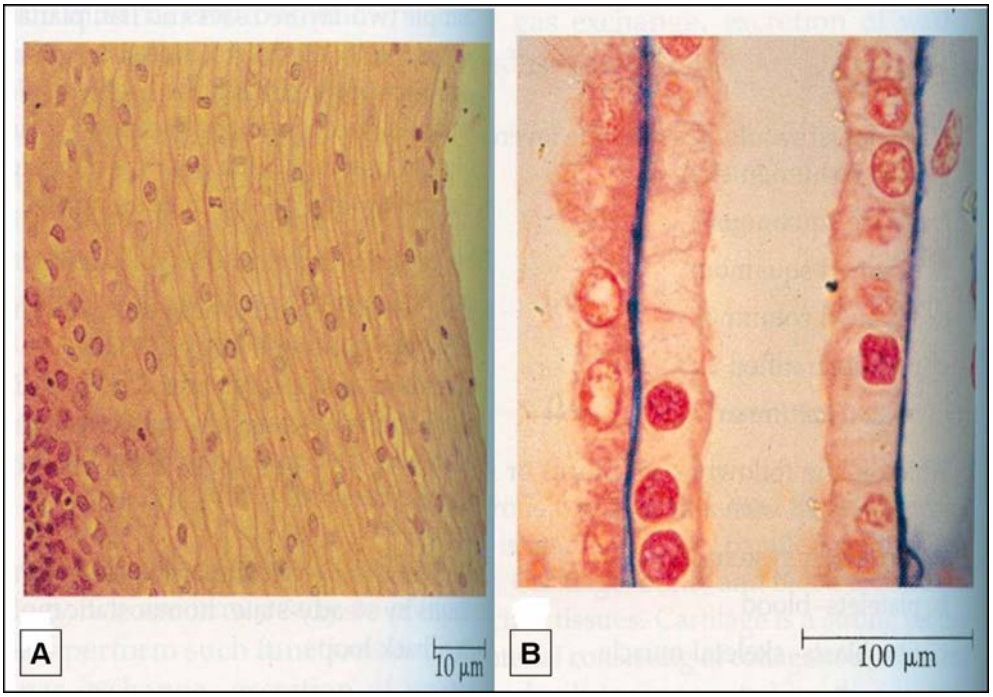


Figure A.43: Two pictures of epidermal tissue. A= stratified squamous epithelium; B= simple cuboical epithelium. Adapted from Campbell 1990, page 792.

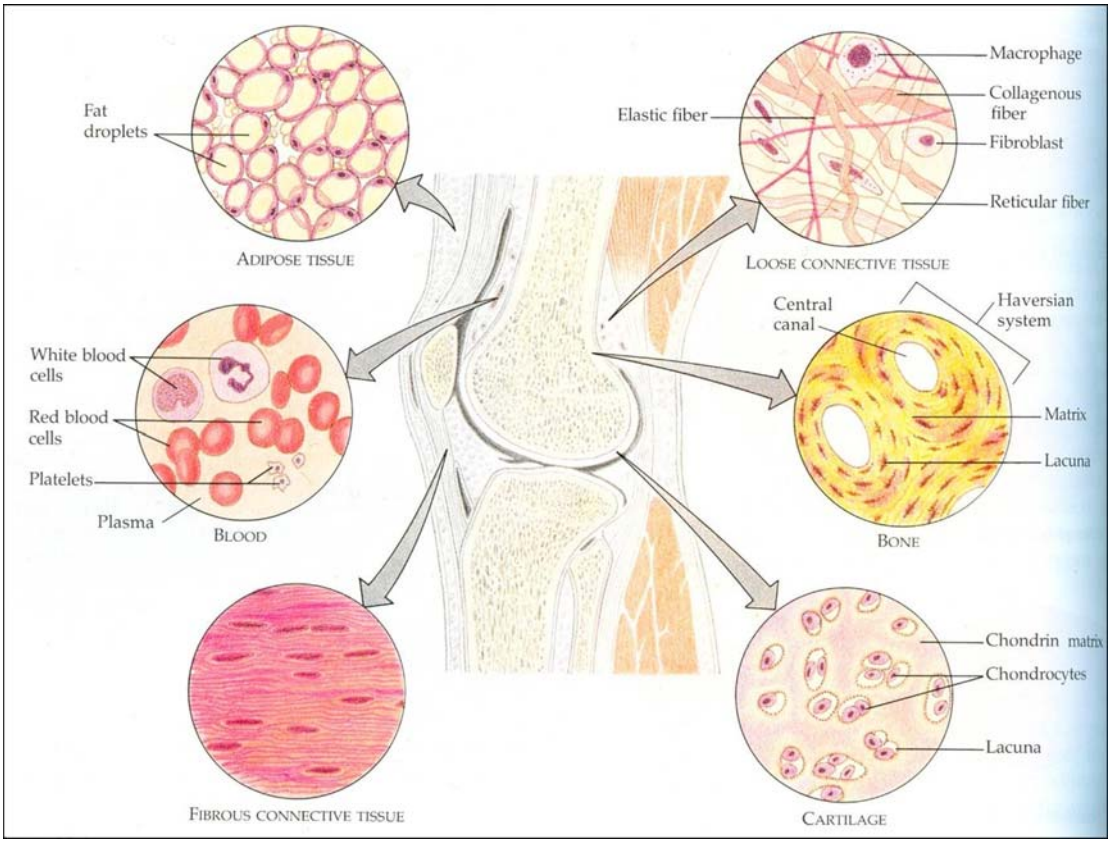


Figure A.44: Some types of connective tissue. The area in the picture is the region around the knee joint. Adapted from Campbell 1990, Figure 36.3, page 784.

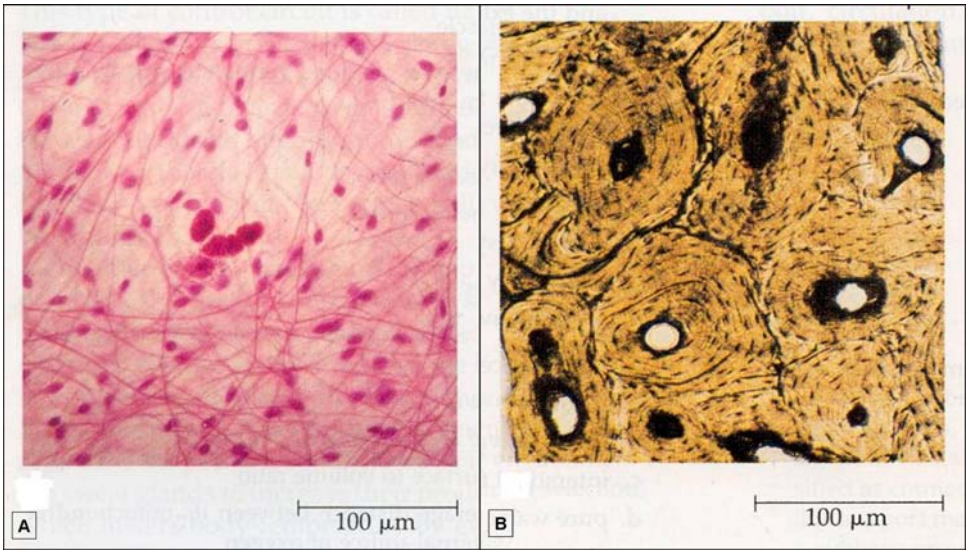


Figure A.45: Two connective tissues. A= loose connective tissue; B= bone. Adapted from Campbell 1990, page 792.

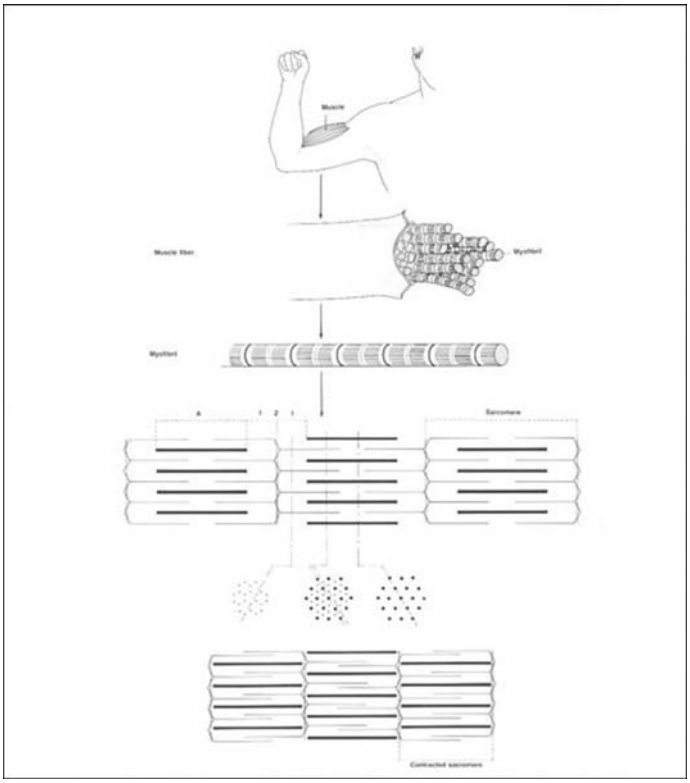


Figure A.46: Drawing of the microanatomy of muscle tissue, showing its levels of organisation from the whole muscle to microfilament. Adapted from Moran & Rowley 2008, plate 7-1.

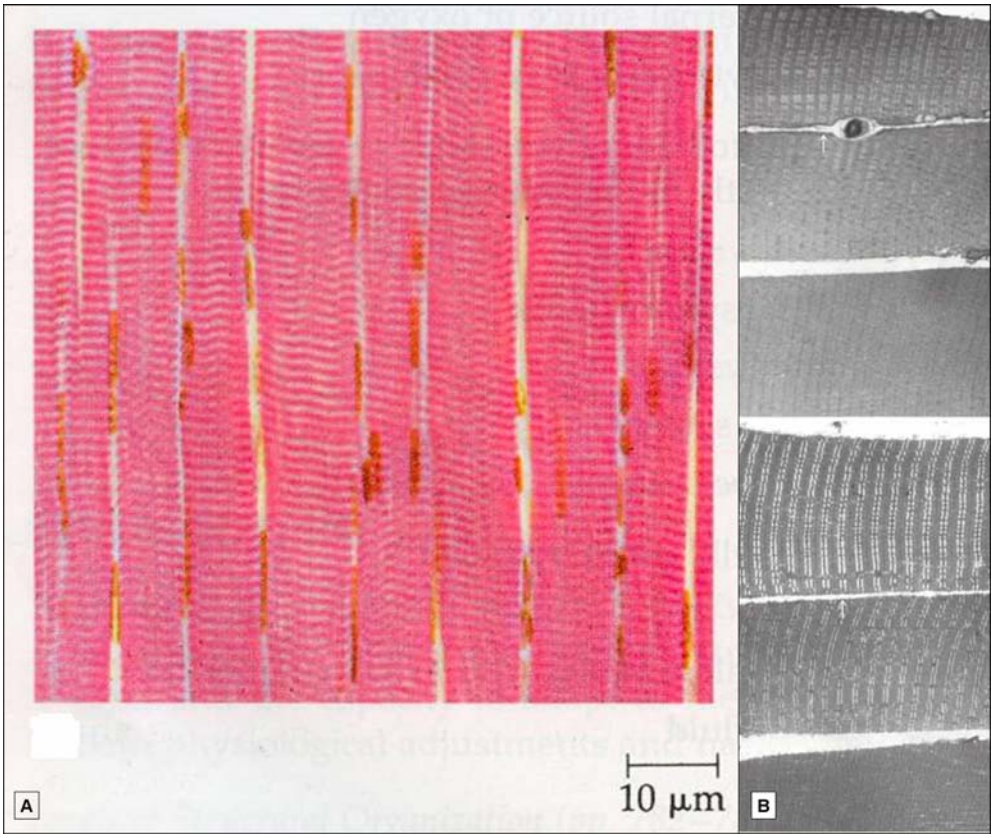


Figure A.47: Several images of striated skeletal muscle tissue. A= adapted from Campbell 1990, page 792; B= SEM image by Moran & Rowley 2008, plate 7-2. Magnification Figure B is 2,800.



Figure A.48: Nervous tissue. Adapted from Campbell 1990, page 792.

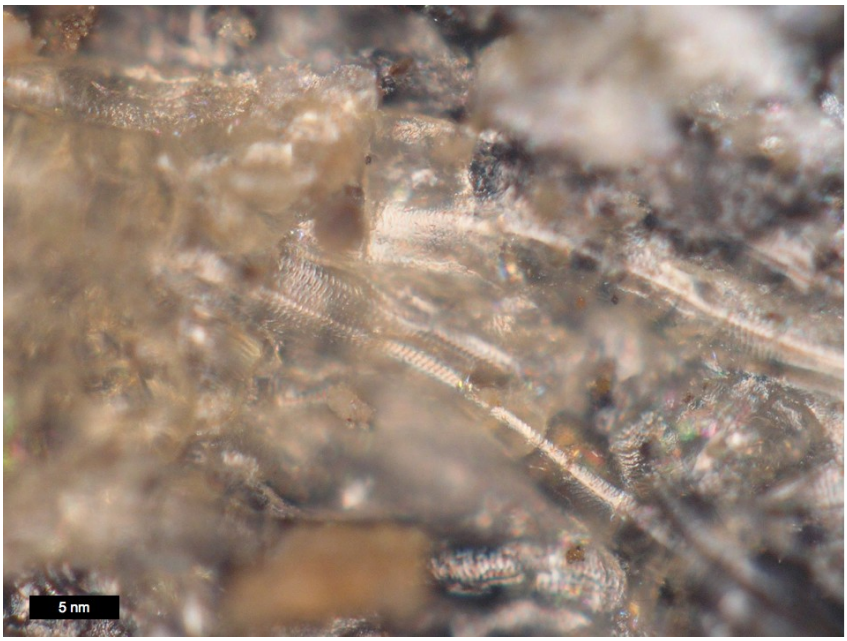


Figure A.49: Micrograph of birefringent striated skeletal muscle tissue. Note the dark and light banding of the sarcomeres. Magnification 200, brightfield cross-polarised light.

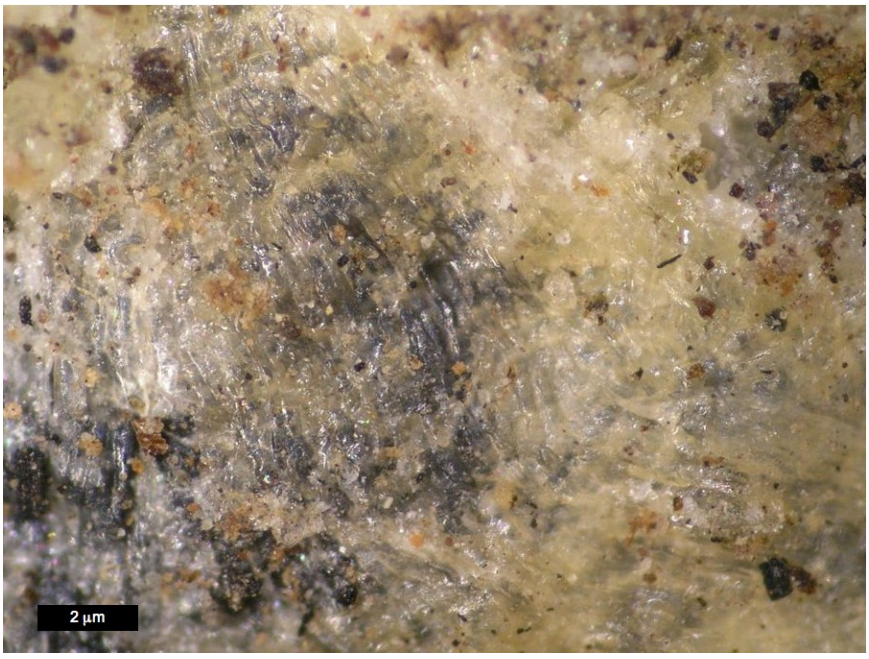


Figure A.50: Micrograph of striated skeletal muscle tissue. Magnification 50.

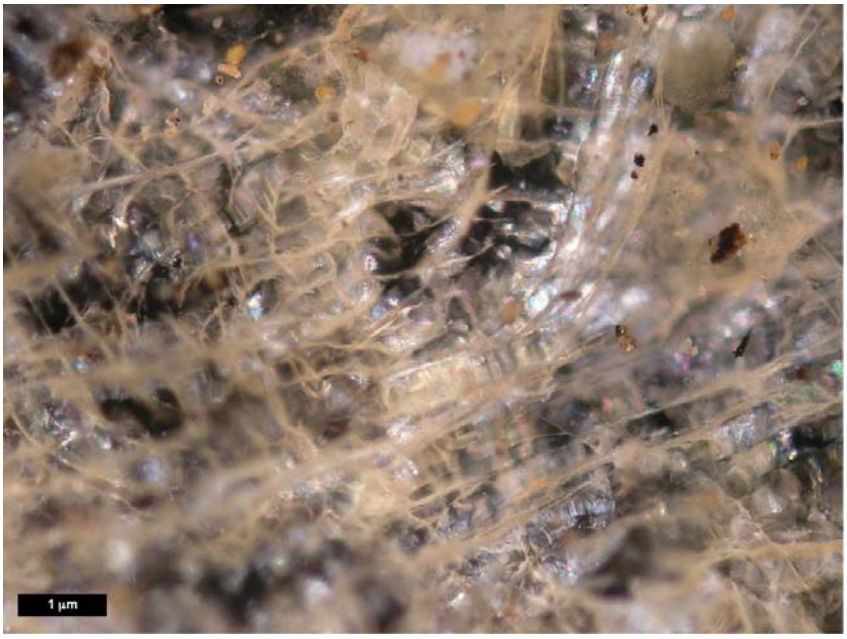


Figure A.51: Micrograph of birefringent striated skeletal muscle tissue. Note that muscle tissue can also be translucent. Magnification 100, brightfield polarised light.

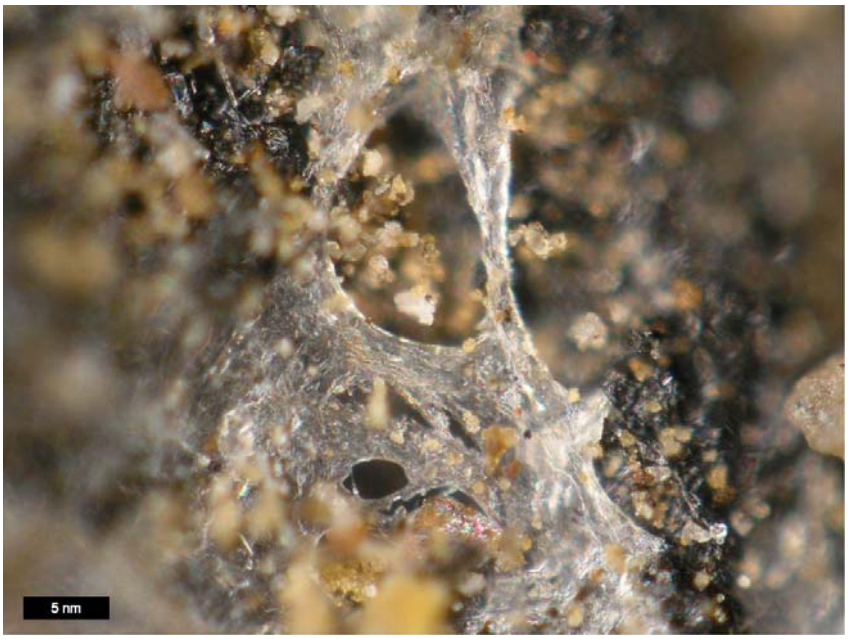


Figure A.52: Micrograph of "gelatinous" muscle tissue. This deposit is similar to the "gelatinous hyphae" in A.37. Magnification 200.



Figure A.53: Micrograph of muscle tissue. Magnification 50.



Figure A.54: Micrograph muscle fibre. Magnification 50.



Figure A.55: Micrograph of muscle fibres. Muscle fibres are more organic and less rigid than plant fibres, see for example A.9. Magnification 50.

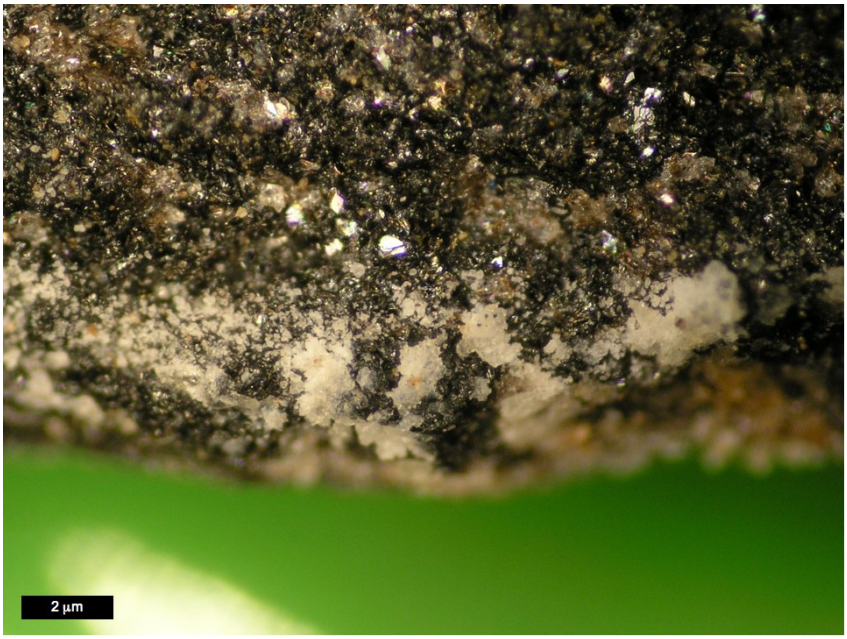


Figure A.56: Micrograph of amorphous fatty bone. Magnification 50.



Figure A.57: Micrograph of bone flakes. Bone and fat are opaque and often have a bluish glow. Magnification 50.

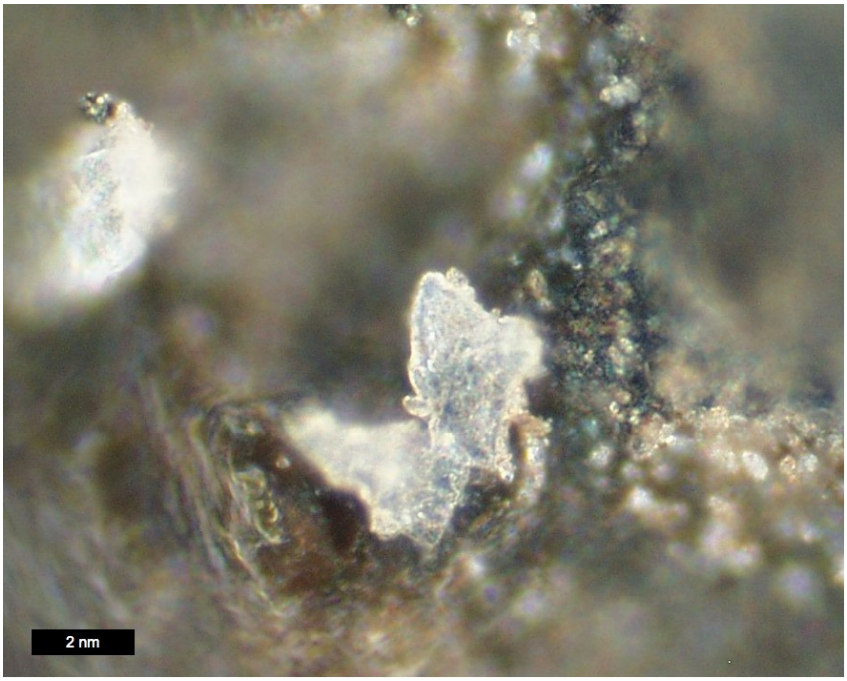


Figure A.58: Micrograph of bluish bone flakes. Magnification 500.

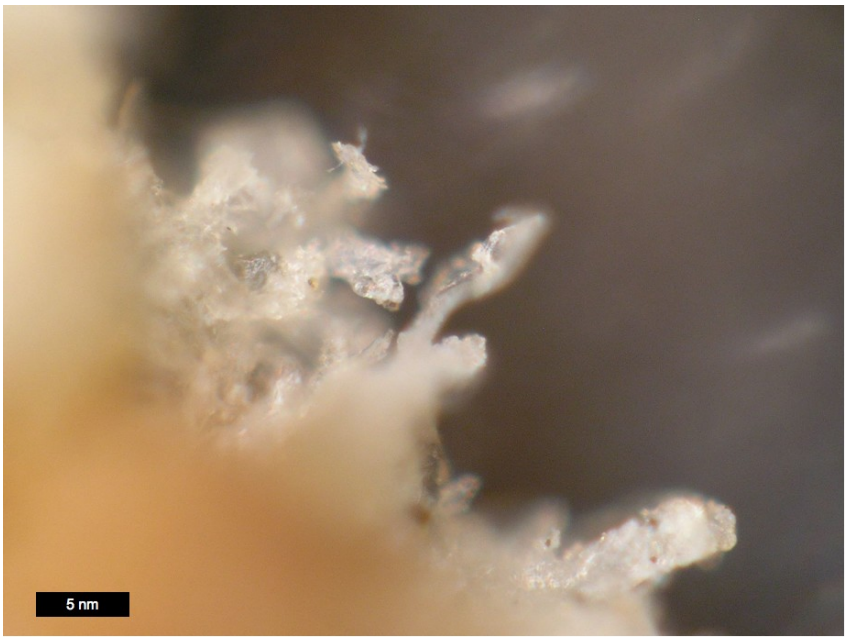


Figure A.59: Micrograph of connective fibres. Connective fibres are opaque and white/bluish. They are more rigid than muscle fibres, which are generally red to yellow in colour and more translucent. Plant fibres are translucent and not bluish. Magnification 200.



Figure A.60: Micrograph of burnt fatty bone. Magnification 100.



Figure A.61: Micrograph of fat cells. Note that the cells have a distinct globular shape. Magnification 50.



Figure A.62: Micrograph of amorphous fat. Magnification 50.



Figure A.63: Micrograph of amorphous fat. Magnification 50.

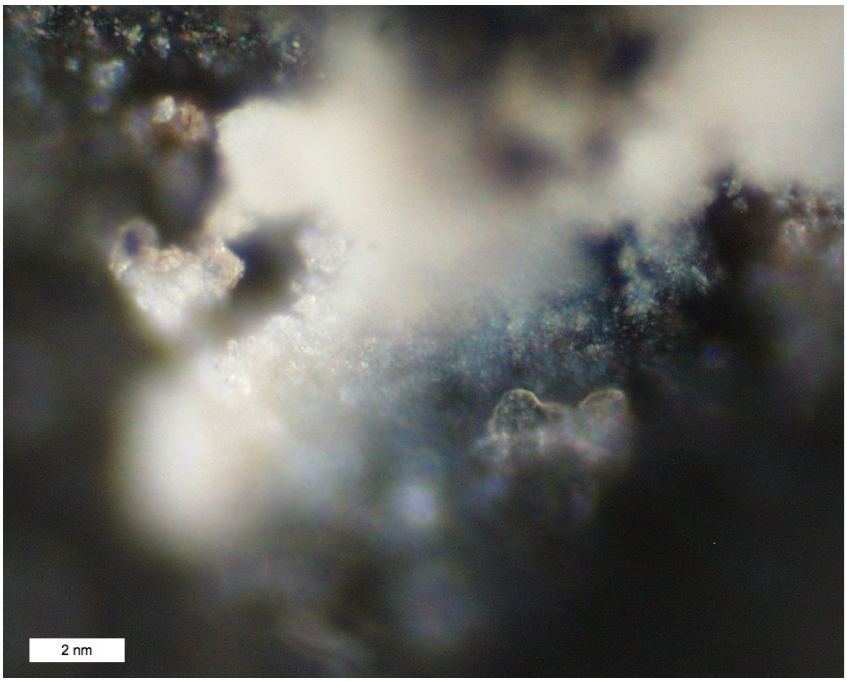


Figure A.64: Micrograph of a bluish fat deposit. Note that this fat deposit has more granular feel than the previous examples. Magnification 500.

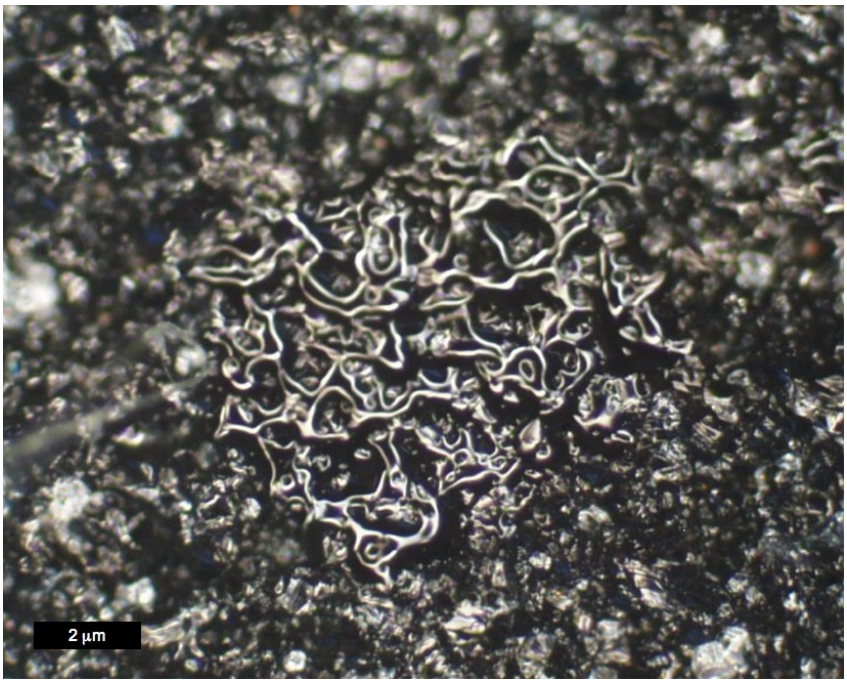


Figure A.65: Micrograph of finger grease. Magnification 50.

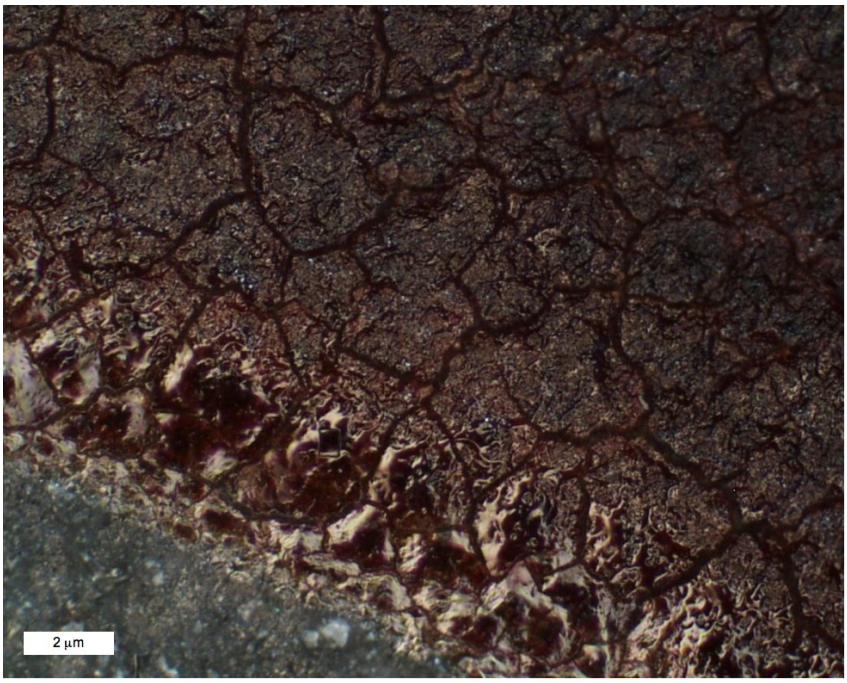


Figure A.66: Micrograph of dried blood. Note the mud cracked surface Magnification 50.

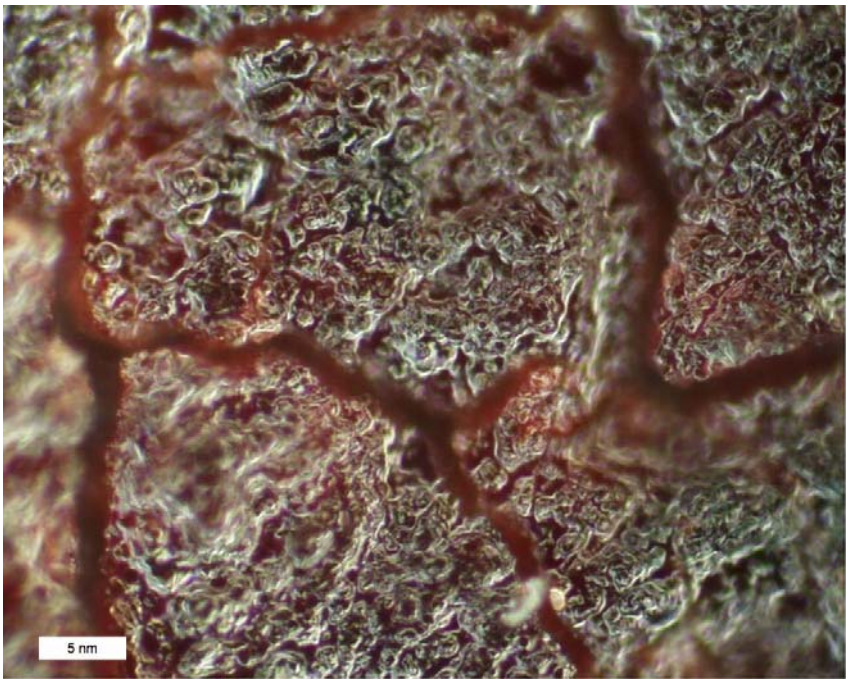


Figure A.67: Micrograph of blood. Note the mud cracked surface. Magnification 200.

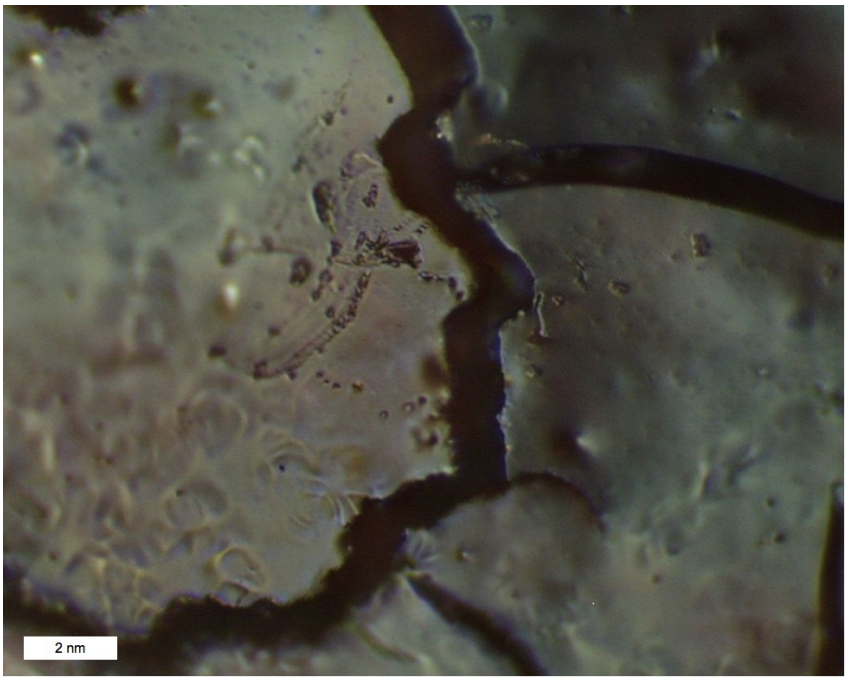


Figure A.68: Micrograph of opaque blood. Magnification 500, brightfield cross-polarised light.

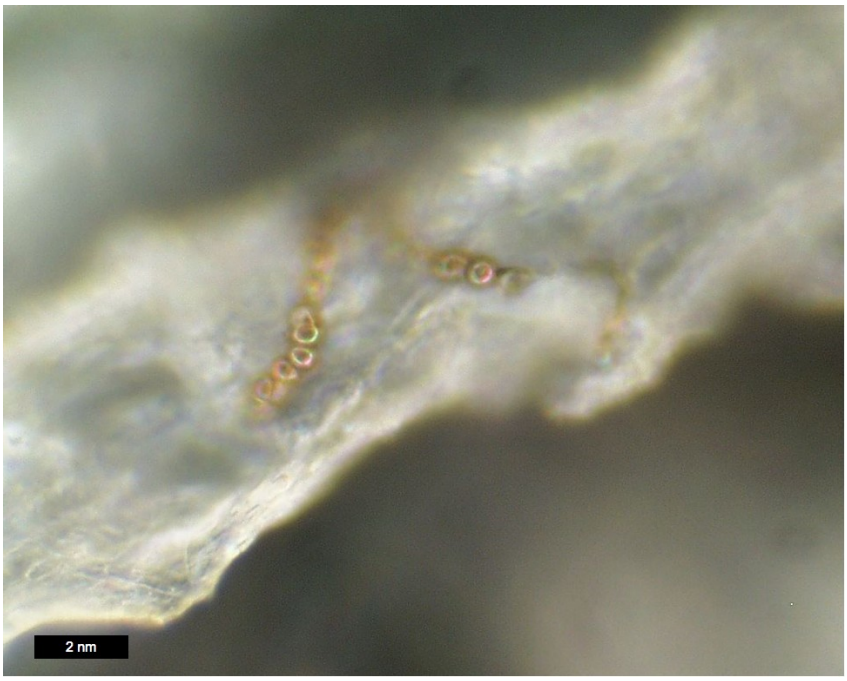


Figure A.69: Micrograph of red blood cells in muscle tissue. Magnification 200.

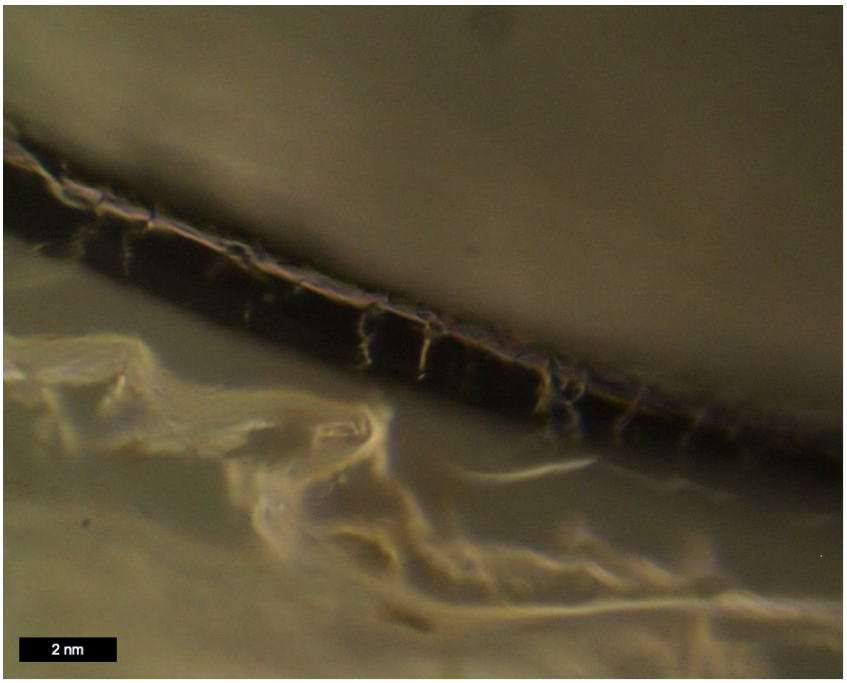


Figure A.70: Micrograph of the scaled surface of hair. Magnification 500.

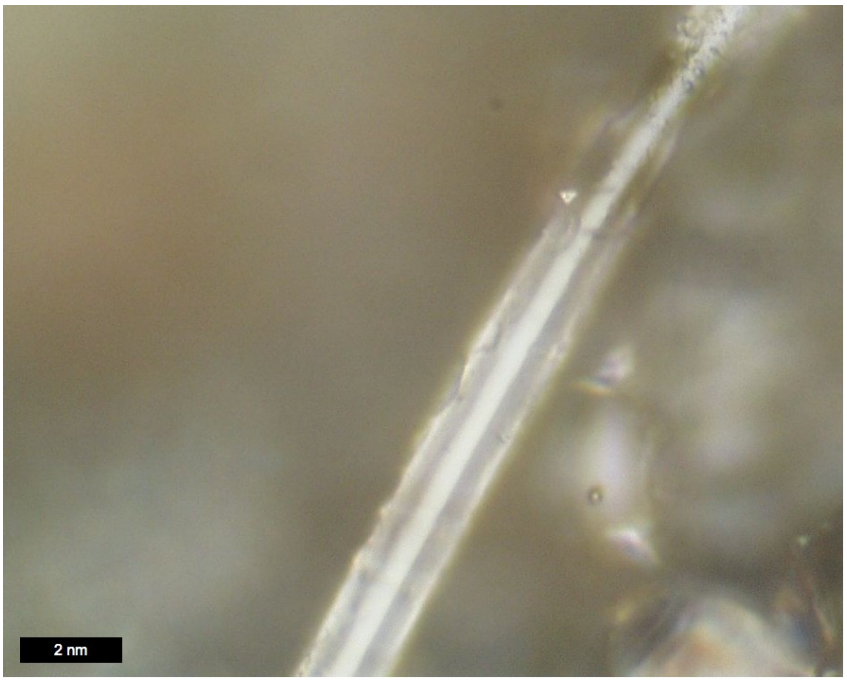


Figure A.71: Micrograph of a medulla in hair. Magnification 200, brightfield cross-polarised light.



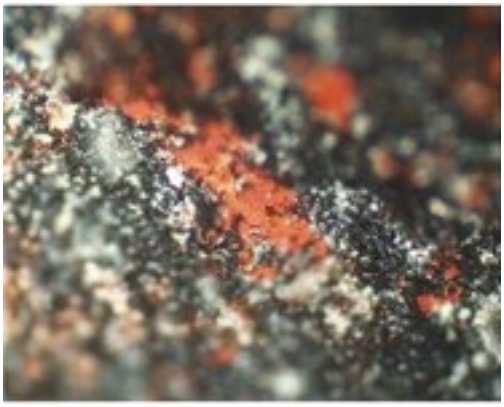
Figure A.72: Micrograph of an insect wing. Magnification 50.



Figure A.73: Micrograph a plastic flake. Unlike bone flakes, plastic flakes are translucent. Magnification 200.



Figure A.74: A replicated and hafted stone tool with resin/gum adhesive and ochre staining on the stone surface (courtesy of Lyn Wadley).


A

B

Figure A.75: Micrographs of modern red ochre on replicated tools. Magnification 50 (courtesy Marlize Lombard).



Figure A.76: Micrograph of white ash. White ash is different from fat deposits like A.64 in that it has clear edges and it is generally not smeared on the surface like fat is. It also lacks the blue haze and is truly granular. Magnification 100.



Figure A.77: Micrograph of white ash. Note the granular character of the deposit. Magnification 200.



Figure A.78: Micrograph of white ash in Sibudu sediment. Magnification 100.

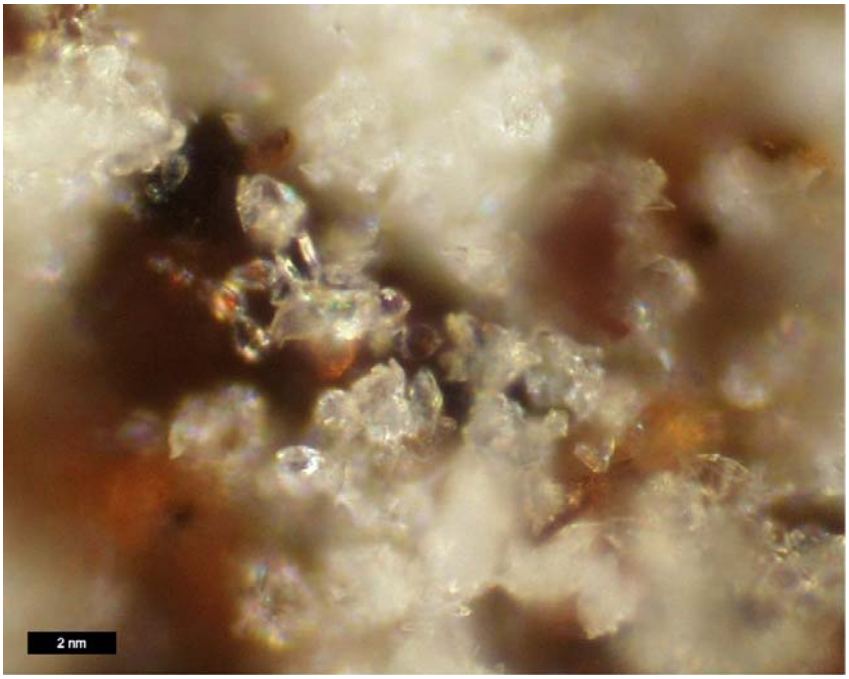


Figure A.79: Micrograph of white ash in Sibudu sediment. Note that the ash grains are more angular and have ragged edges. This is different from the more circular and smooth starch grains. Magnification 500, brightfield polarised light.

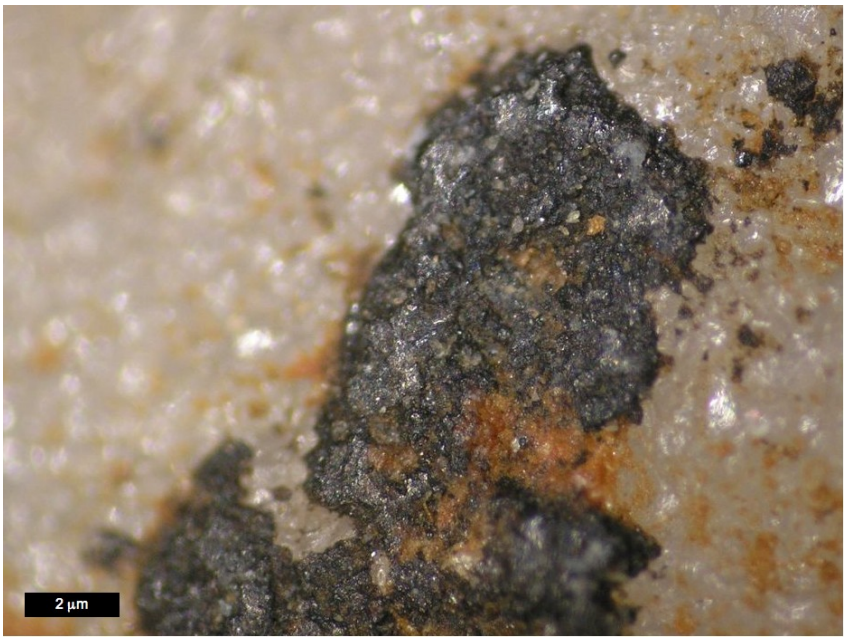


Figure A.80: Micrograph of black deposits, probably manganese. Magnification 50. Photo taken from archaeological sample STK2053*.

* There are no modern or replicated examples of black manganese deposits, however similar deposits are present on the breccia samples, see for example FigureA.104.

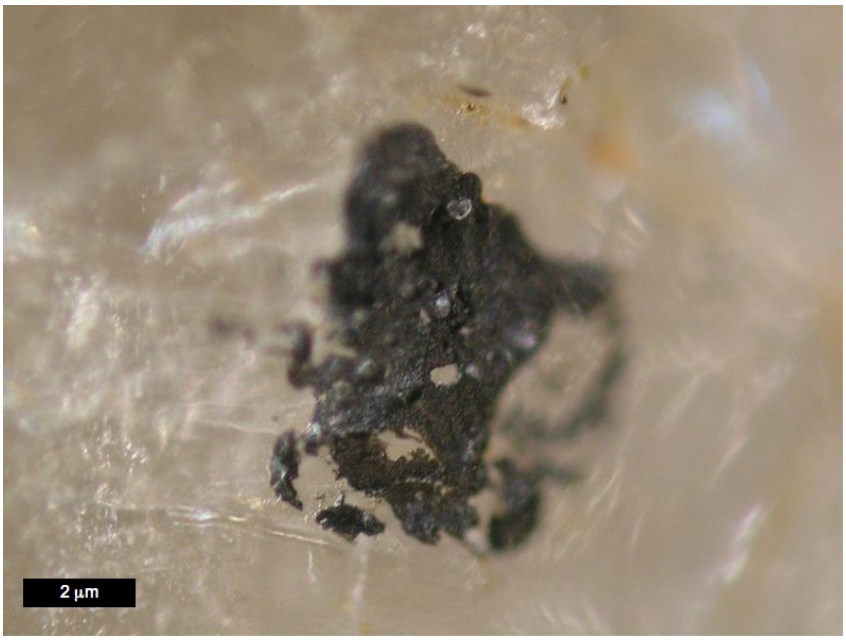


Figure A.81: Micrograph of a black deposit, probably manganese. Magnification 50. Photo taken from archaeological sample STK6048*.

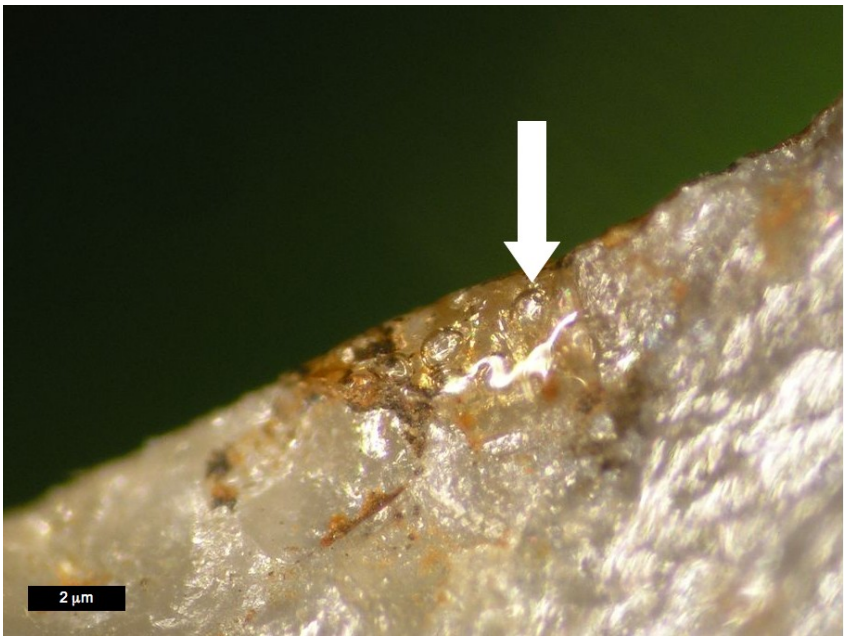


Figure A.82: Micrograph of translucent nail varnish with two air bubbles (indicated by the arrow). Nail varnish is, unlike resin, extremely clear and translucent. In this example it also overlies a thin sediment deposits, indicating that the varnish was applied after excavation. Magnification 50.

* There are no modern or replicated examples of black manganese deposits, however similar deposits are present on the breccia samples, see for example FigureA.104.

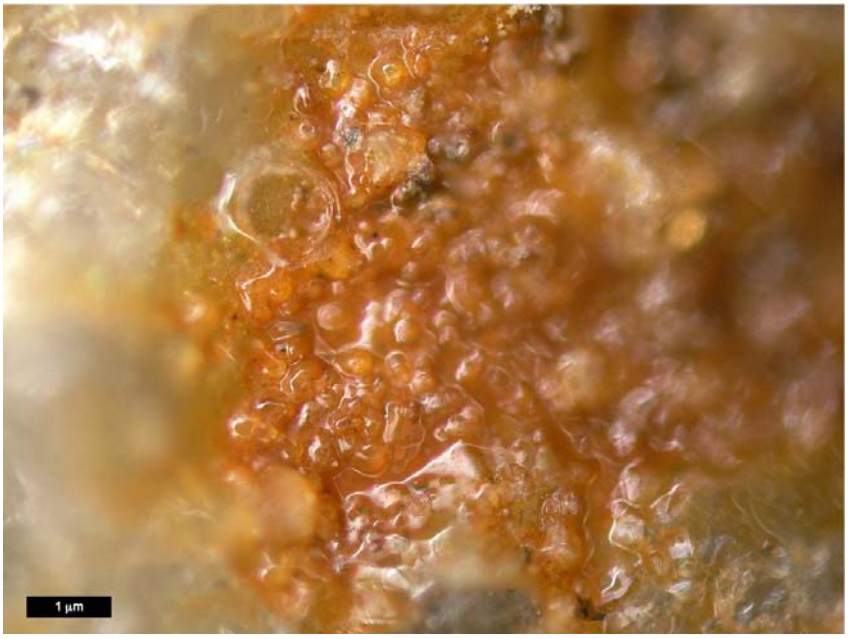


Figure A.83: Micrograph of sediment under nail varnish. Magnification 100.

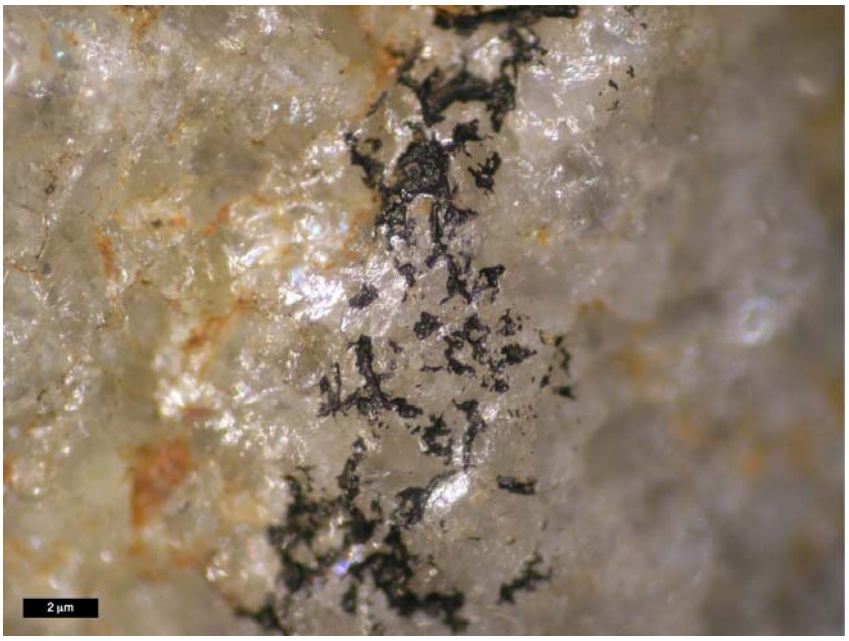


Figure A.84: Micrograph of ink. Magnification 50. Photo taken from archaeological sample STK6085*.

* Although the examples of ink do not derive from a modern comparative sample, I am sure that these deposits are ink because they form letters on the tool's surfaces, similar to the writing in Figure C.93.

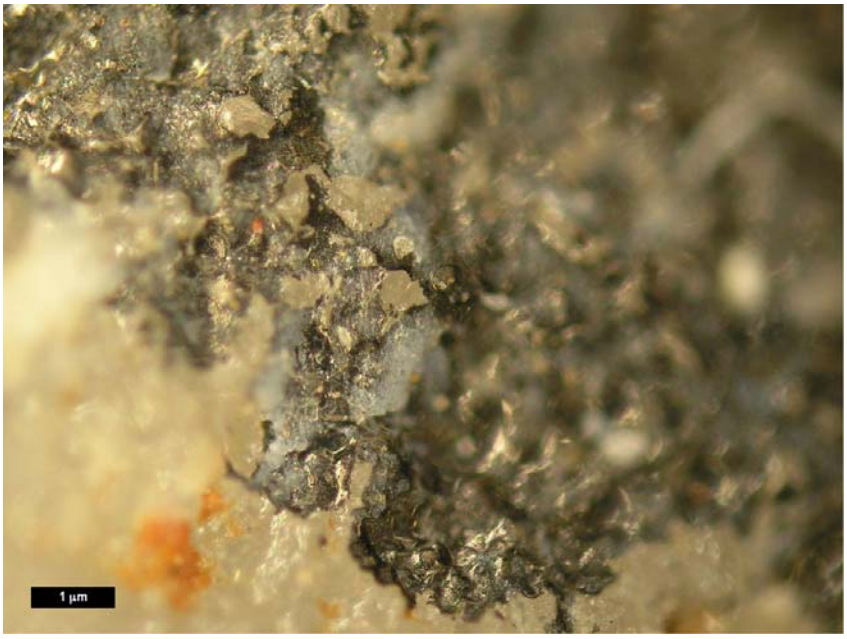


Figure A.85: Micrograph of ink. Magnification 100. Photo taken from archaeological sample STK2053*.



Figure A.86: Micrograph of a fat-like deposit on a blank sample. Note the blue haze, which is characteristic for fat. Magnification 50.

* Although the examples of ink do not derive from a modern comparative sample, I am sure that these deposits are ink because they form letters on the tool's surfaces, similar to the writing in Figure C.93.

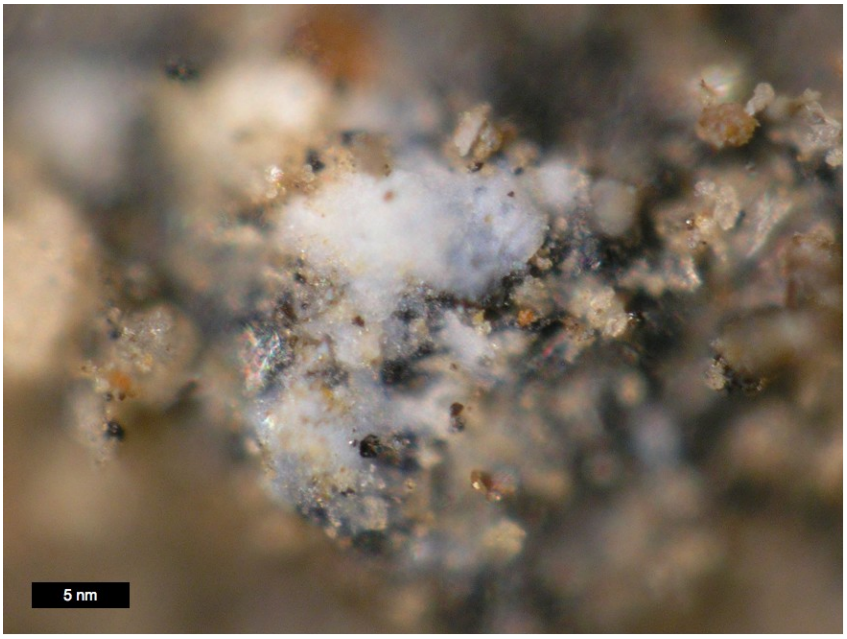


Figure A.87: Micrograph of a fat-like deposit on a blank sample. Note the blue haze, which is characteristic for fat deposits. Magnification 200.

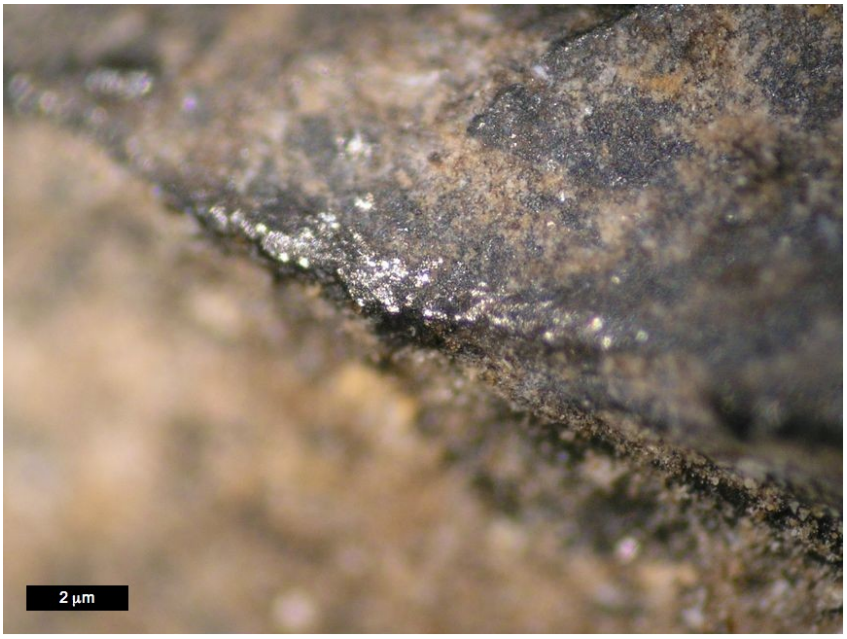


Figure A.88: Micrograph of reflective polish on a ridge. Magnification 50. Photo taken from archaeological sample SBL10.



Figure A.89: Micrograph of reflective polish on an edge. Magnification 50. Photo taken from archaeological sample SBL28.



Figure A.90: Micrograph of broken flint. Note that the flakes are similar to bone flakes. The only way to distinguish them is to apply the contextual approach and note wear and breaking on a tool. Magnification 50.

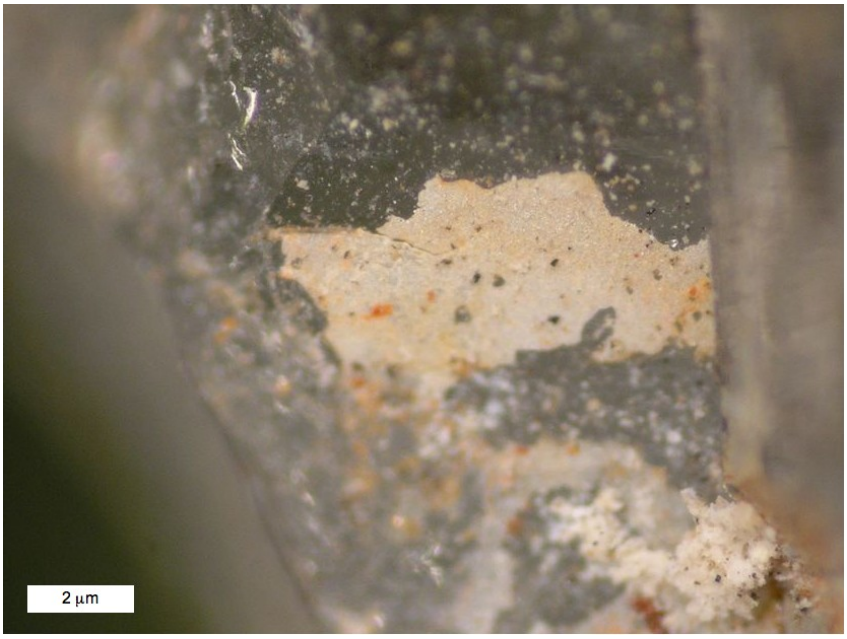


Figure A.91: Micrograph of broken chert. Note that this break resembles bone. Magnification 50.

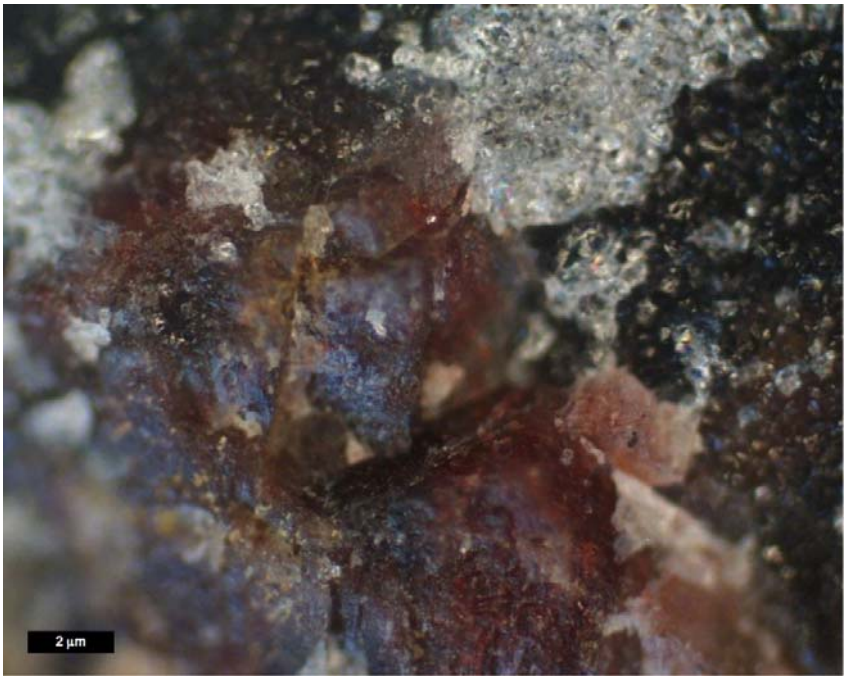


Figure A.92: Micrograph of damaged chert surface with a red inclusion. Magnification 50.

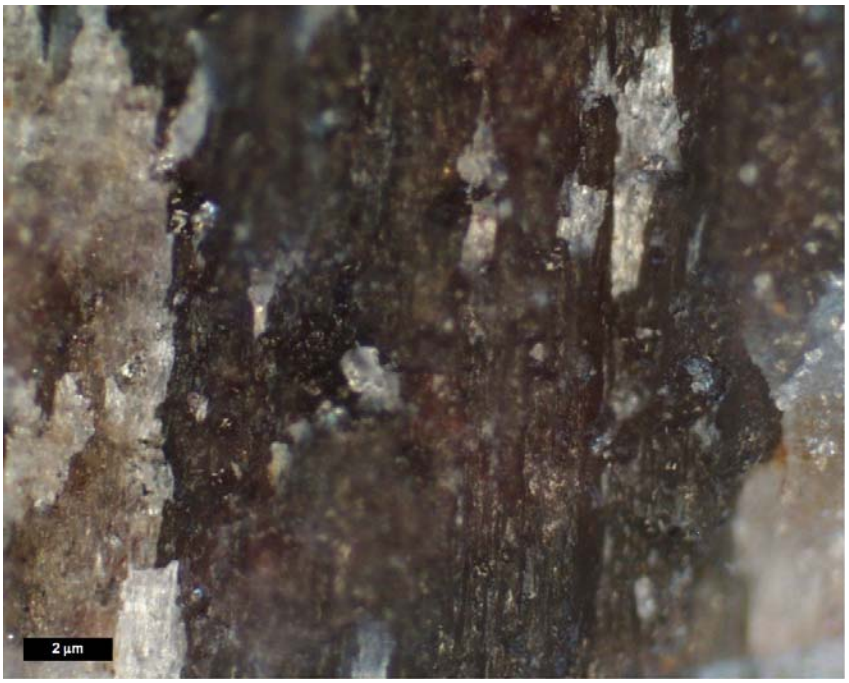


Figure A.93: Micrograph of damaged chert surface with inclusion. Magnification 50.

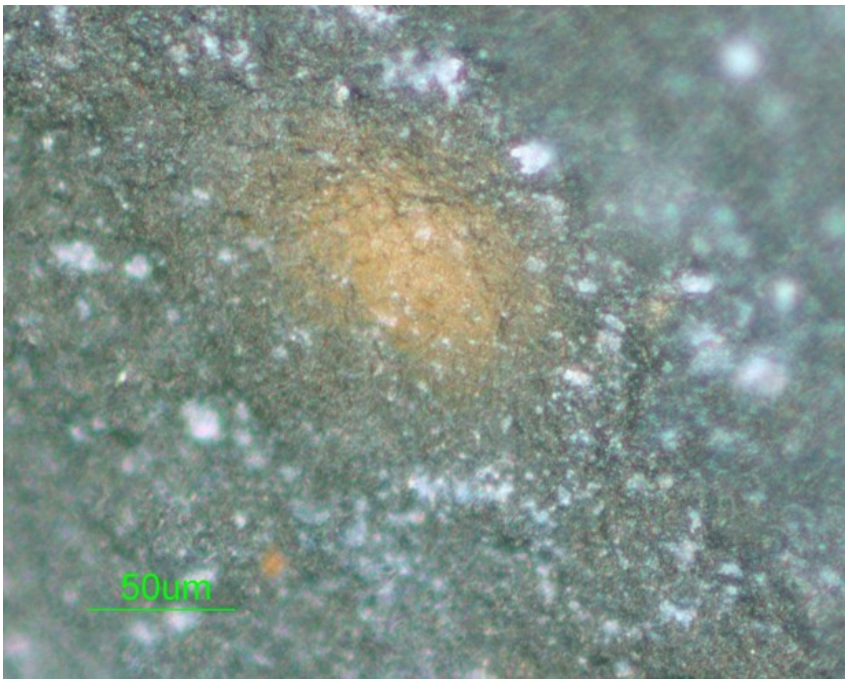


Figure A.94: Micrograph of an inclusion in flint. Magnification 200.

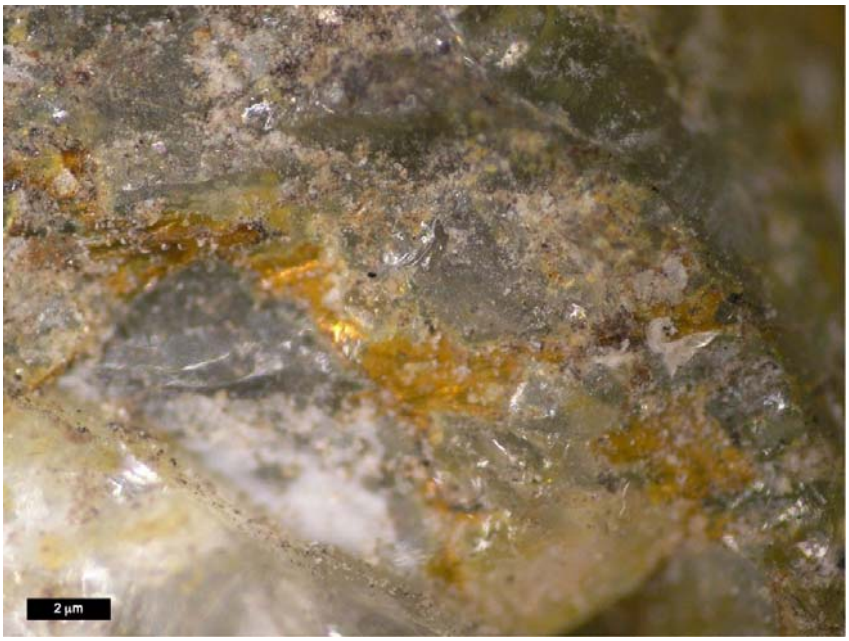


Figure A.95: Micrograph of damaged quartz surface with inclusions. This yellow inclusion resembles a dried liquid deposit, but the edges of this inclusion disappear in the stone and not distinct as with a dried liquid. Magnification 50.

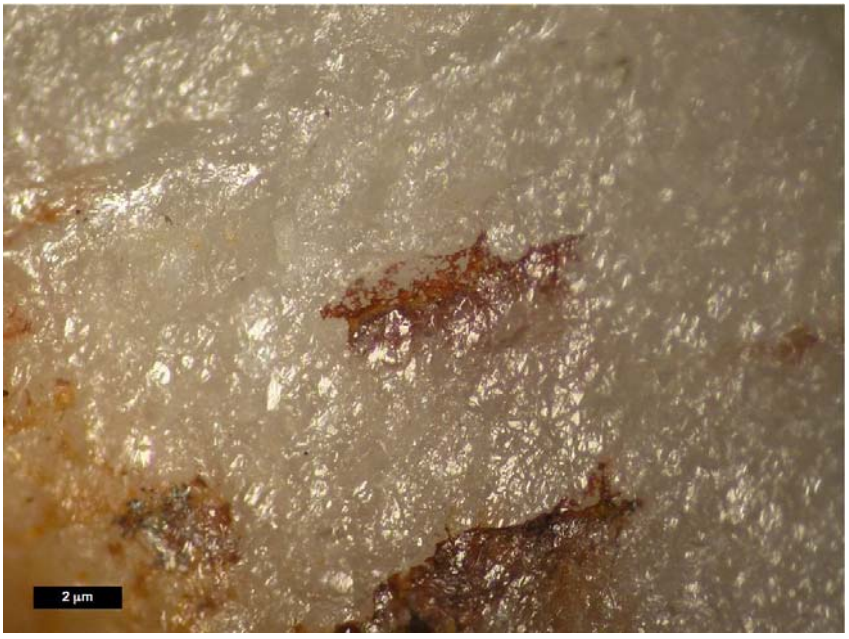


Figure A.96: Micrograph of inclusions in quartz. Note how the inclusions seem to come from deeper in the stone and surface as red deposits. Magnification 50.



Figure A.97: Plant tissue in Sterkfontein sediment. Magnification 50.

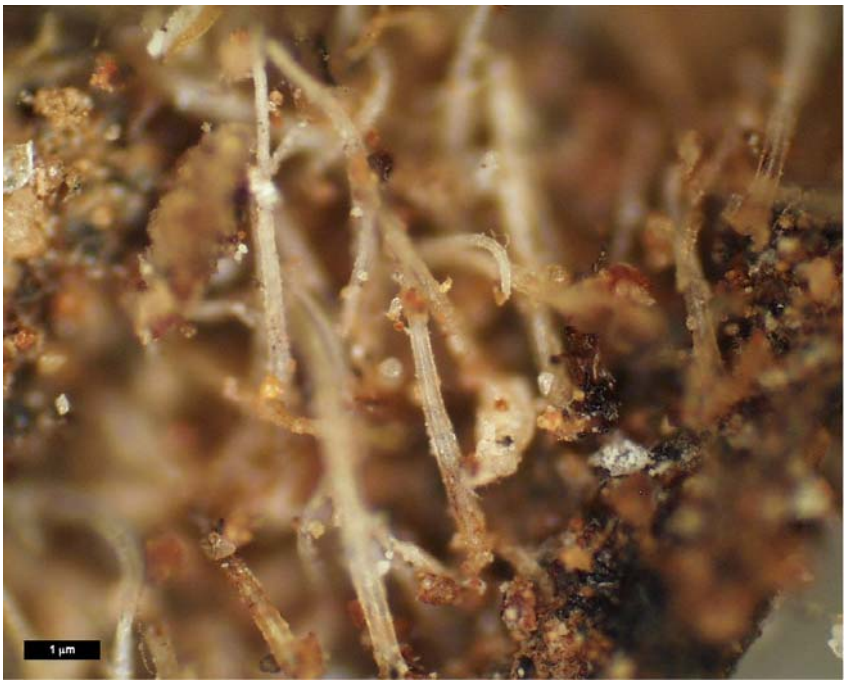


Figure A.98: Rootlets in Sterkfontein sediment. Magnification 100.

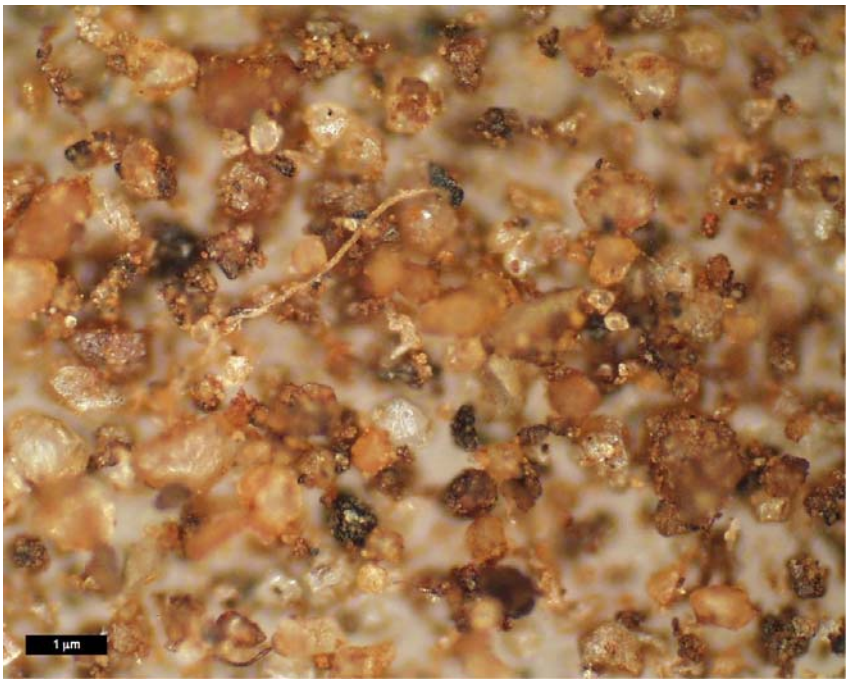


Figure A.99: Sterkfontein sediment matrix. Magnification 100.

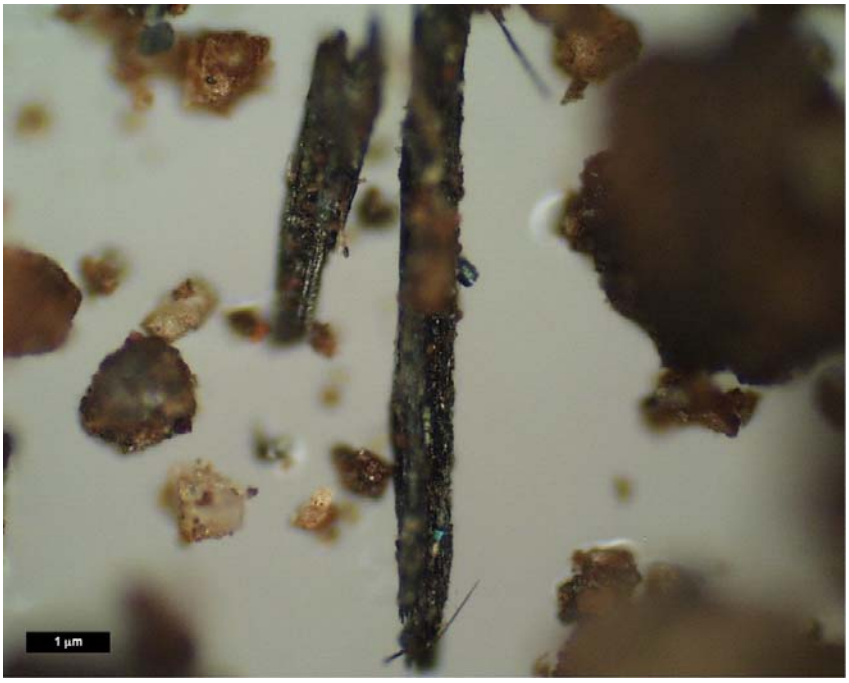


Figure A.100: Charcoal in Sterkfontein sediment. Magnification 100.



Figure A.101: Bone-like particle in Sterkfontein sediment. Note the opaque and bluish properties, which are similar to fat and bone. Magnification 200.

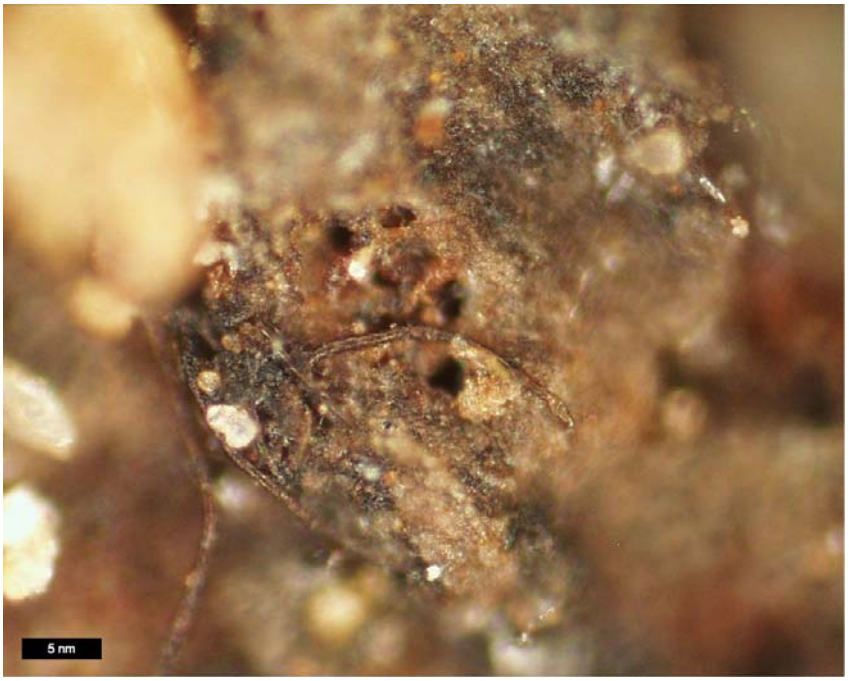


Figure A.102: Hyphae and sediment organics in Sterkfontein sediment matrix. Magnification 200.

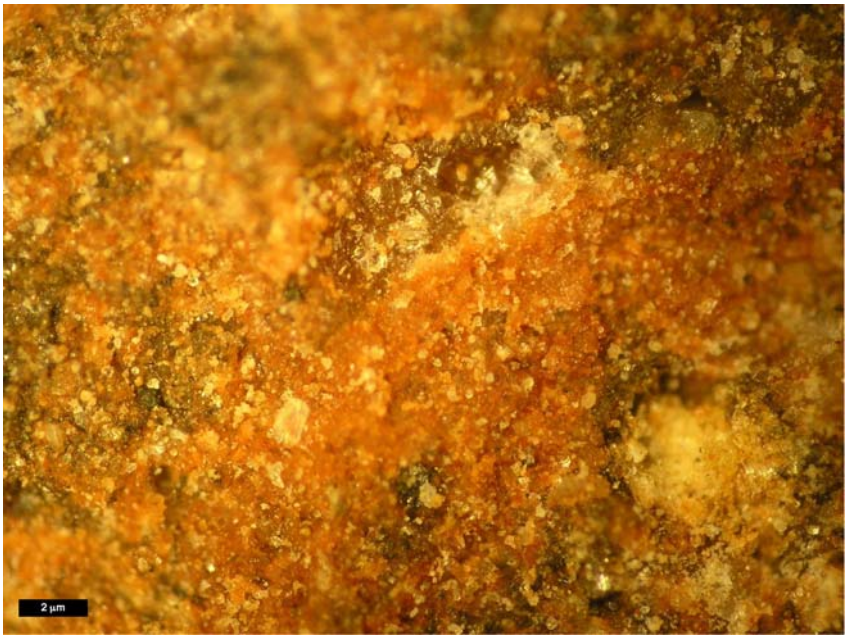


Figure A.103: Orange breccia surface from Sterkfontein. Magnification 50.

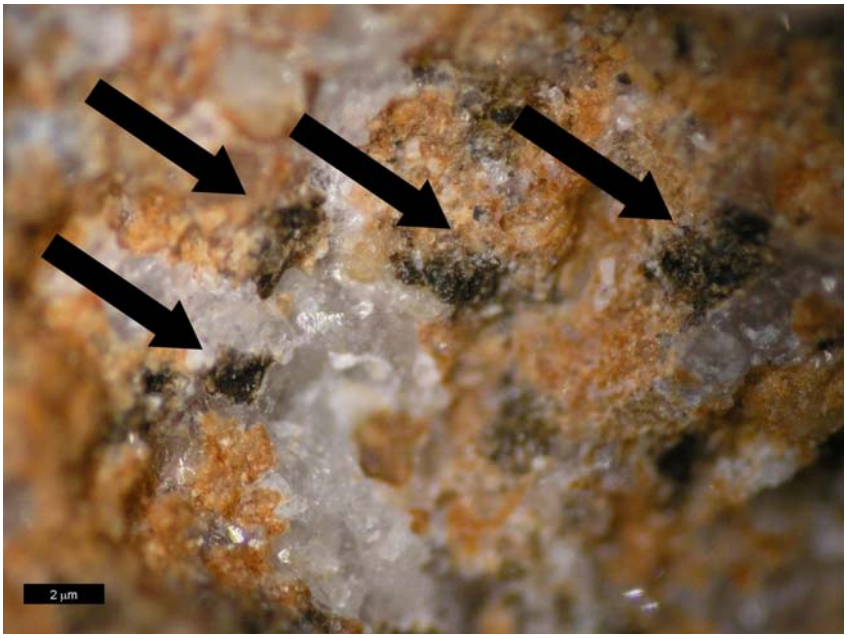


Figure A.104: Breccia surface with white chert clasts and the arrow indicates "black manganese spots". Magnification 50.

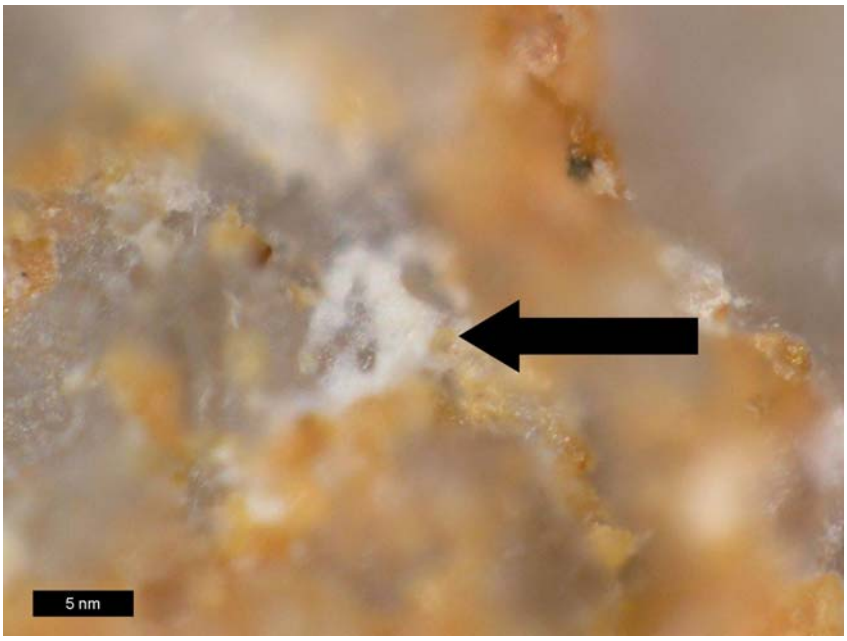


Figure A.105: Arrow indicates fat-like deposit on Sterkfontein breccia. Magnification 200.

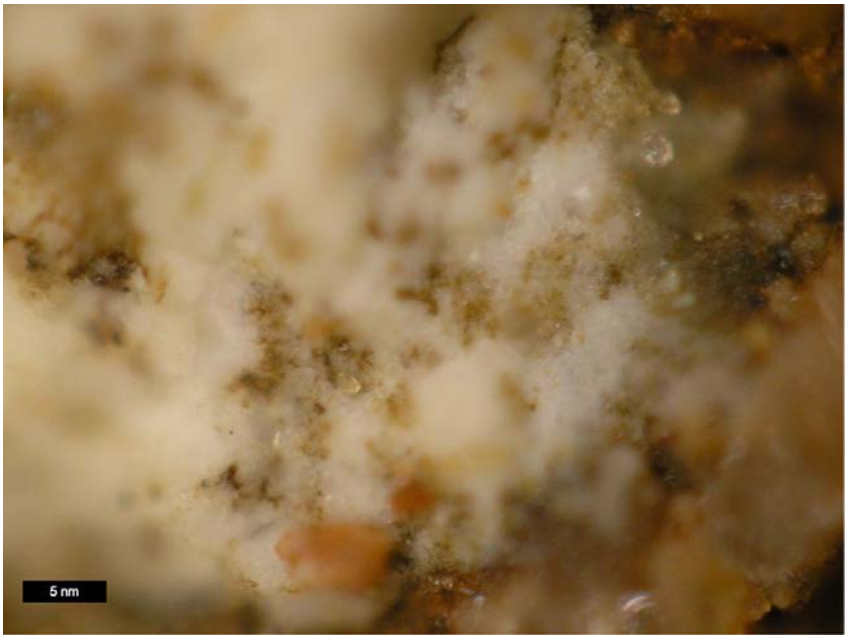


Figure A.106: Fat-like deposit on Sterkfontein breccia. Magnification 200.

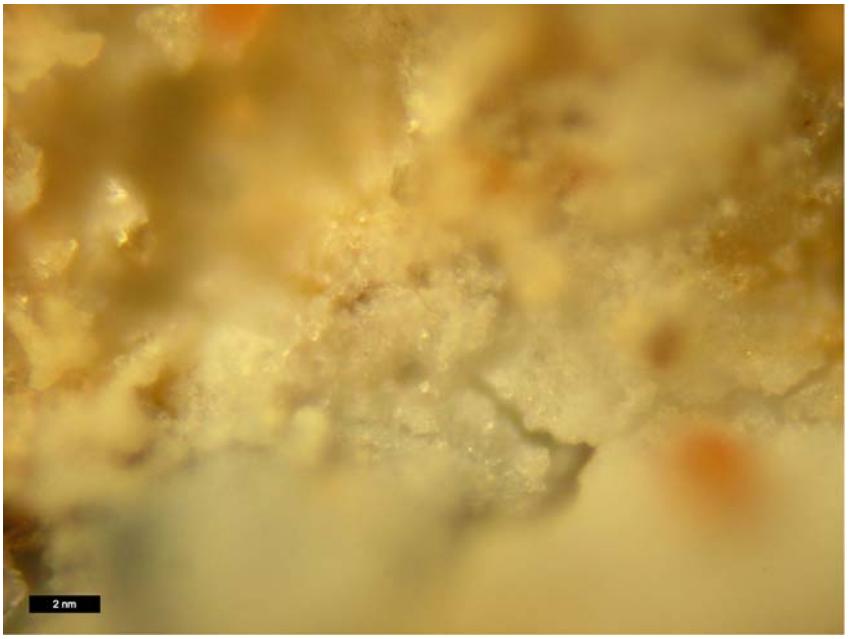


Figure A.107: Fat-like deposit on Sterkfontein breccia. Magnification 500.

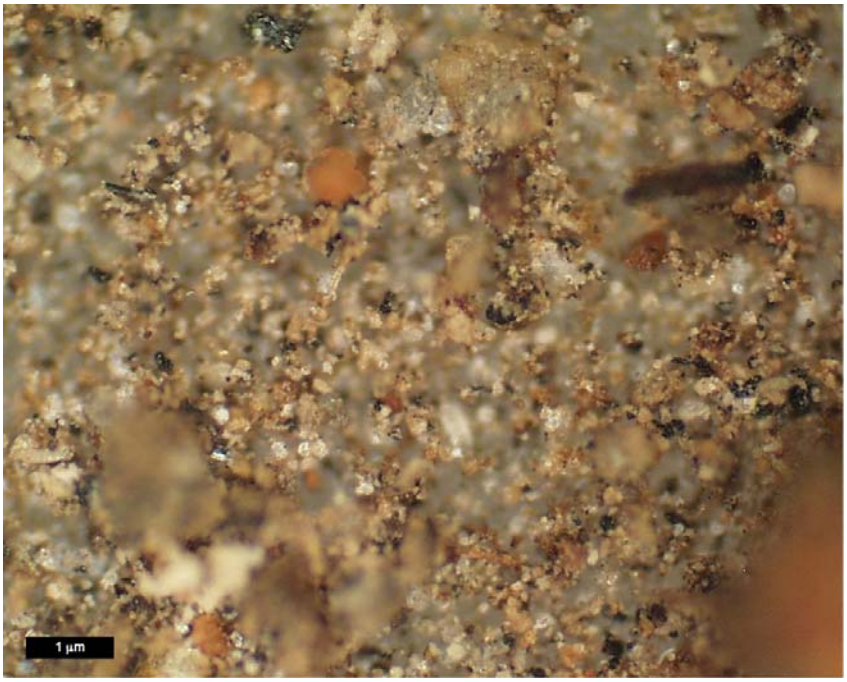


Figure A.108: Sediment matrix of Sibudu. Magnification 100.

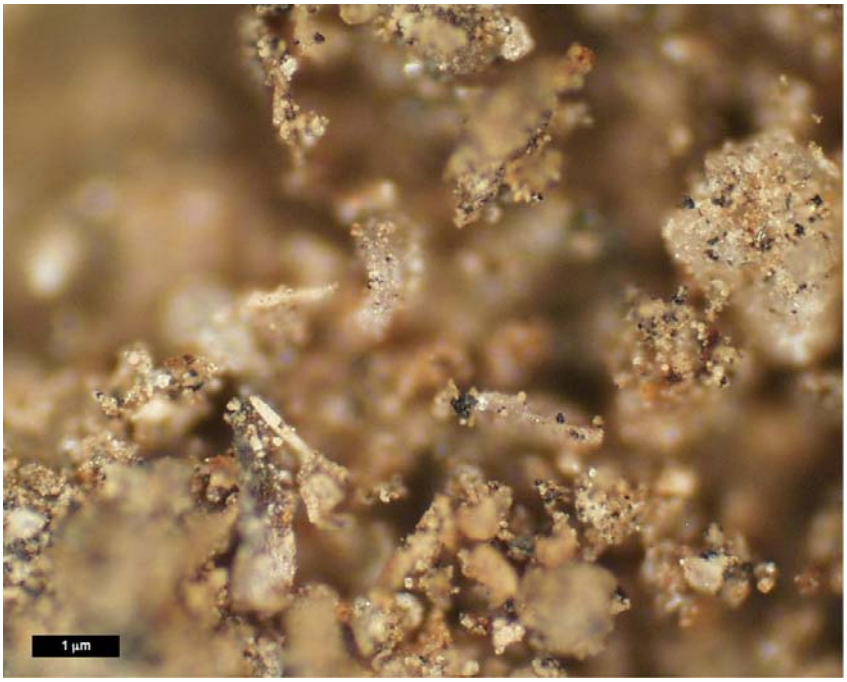


Figure A.109: Sediment organics and ash the in Sibudu sediment matrix. Magnification 100.

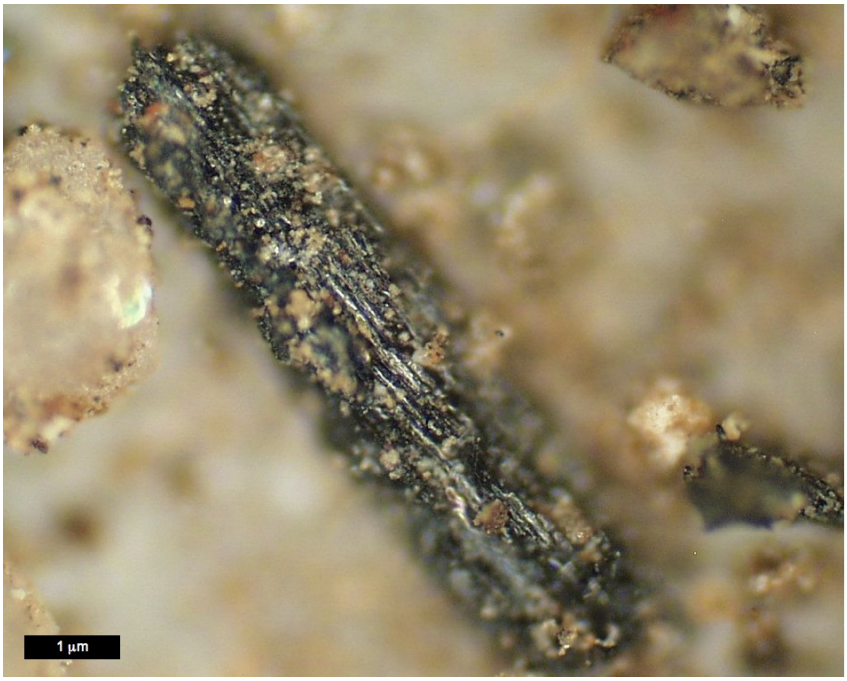


Figure A.110: Charcoal in Sibudu sediment. Magnification 100.



Figure A.111: Brown hyphae in Sibudu sediment. Magnification 100.

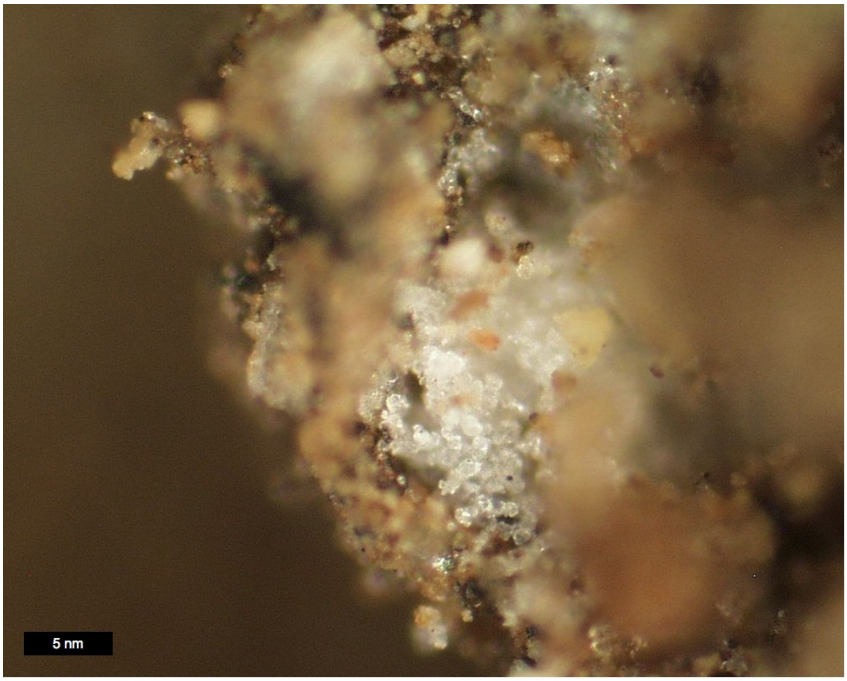


Figure A.112: White spores in Sibudu sediment. Most spores I found are black, but they can be white too. Magnification 200.



Figure A.113: Large rootlet in Sibudu sediment. Magnification 50.

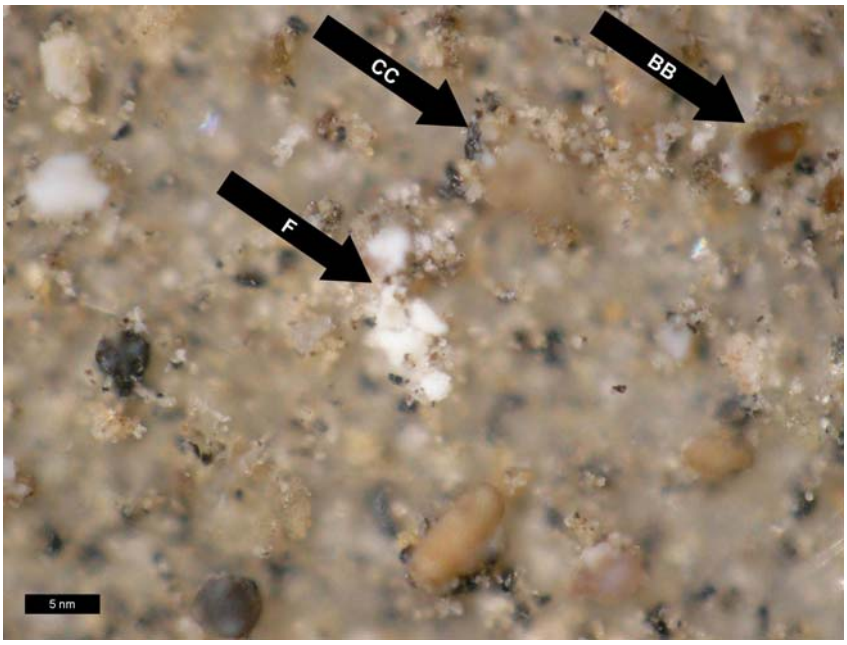


Figure A.114: Sediment matrix in Sibudu layer Grey Rocky II. Note the black charcoal particles, the white fat-like remains and the yellow/orange "burnt bone" particles. BB= burnt bone; CC= charcoal; F= fat-like. Magnification 200.

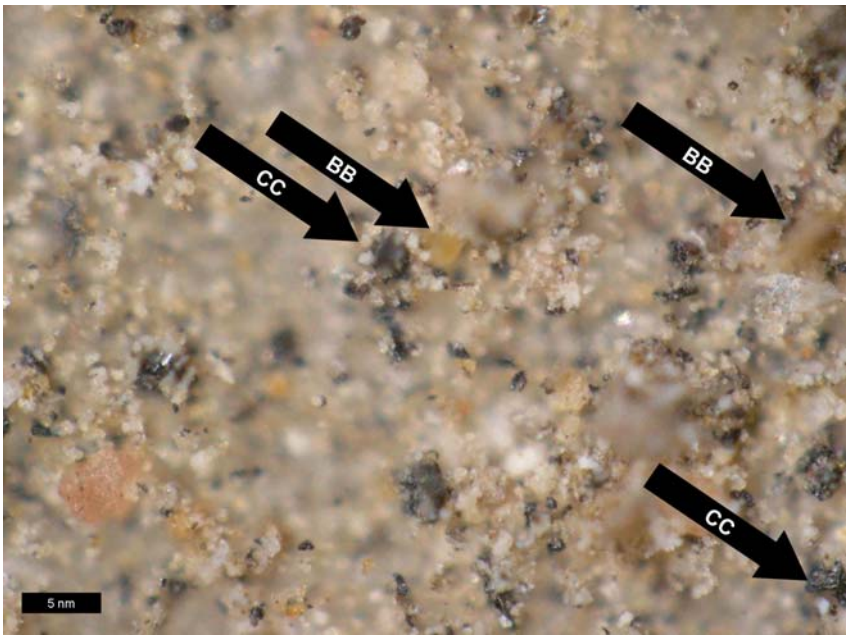


Figure A.115: Sediment matrix in Sibudu layer Grey Rocky II. Note the black charcoal particles and the yellow/orange "burnt bone" particles. BB= burnt bone; CC= charcoal. Magnification 200.

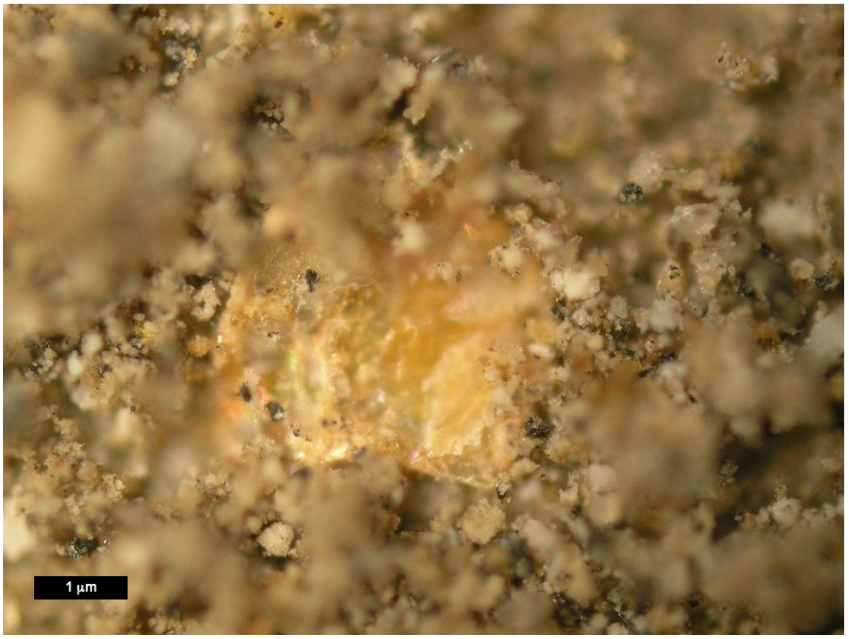


Figure A.116: Yellow/orange "burnt bone" residue in Sibudu layer Grey Sandy II. Magnification 100.



Figure A.117: Orange "burnt bone" residue in Sibudu layer Grey Sandy II. Note that this is similar to A.60 and A.116. Magnification 200.



Figure A.118: Clay from Wilhelmina Polder on flint. Note the fatty character of the flint and clay. Magnification 50.

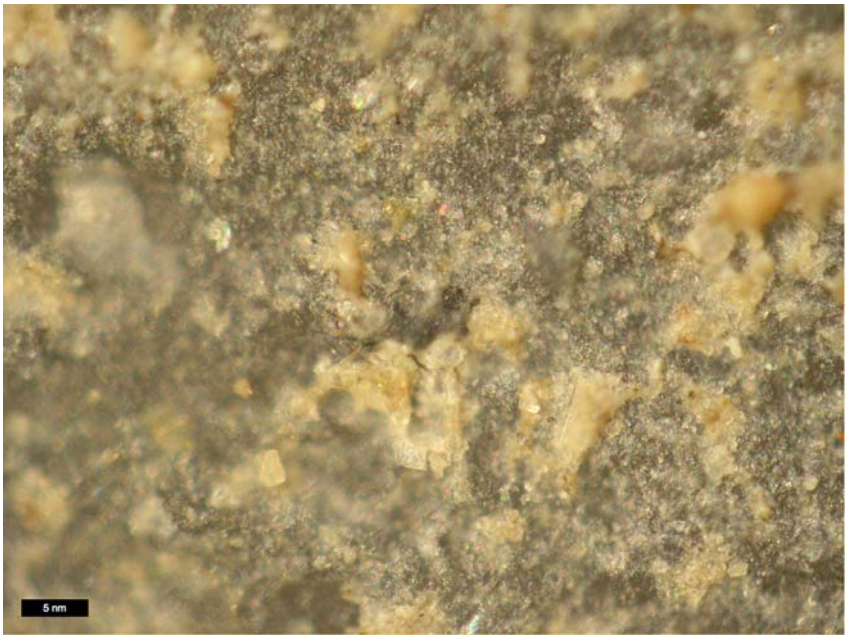


Figure A.119: Clay from Wilhelmina Polder on flint. Magnification 200.

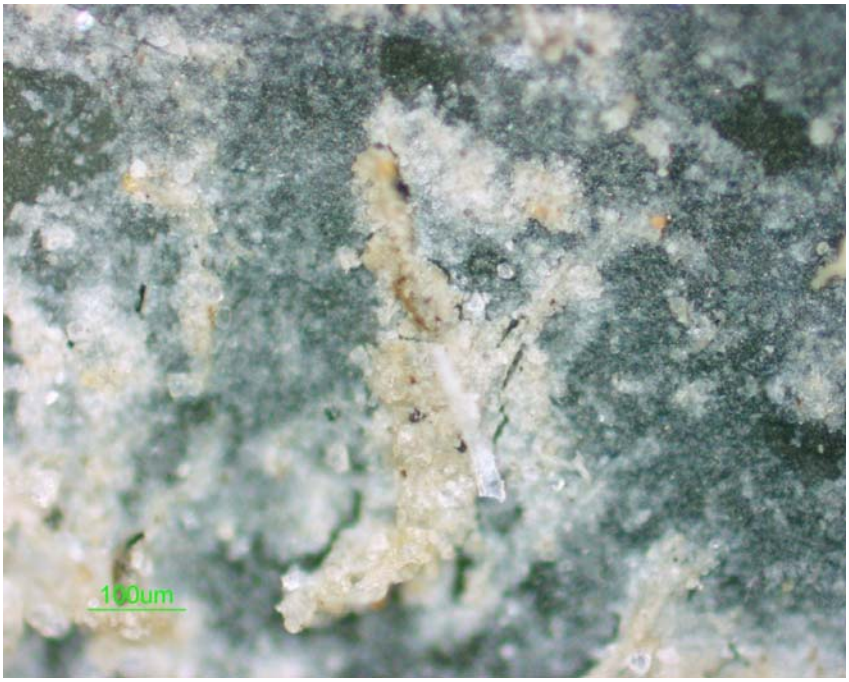


Figure A.120: Clay and a plant fibre in Wilhelmina Polder sediment. Magnification 100.

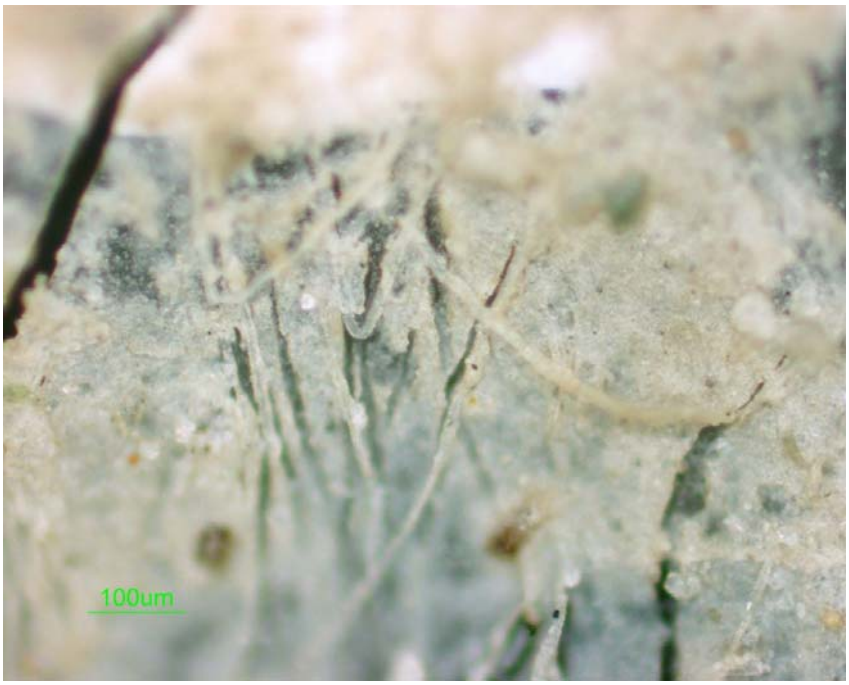


Figure A.121: Clay and plant fibres in Wilhelmina Polder clay sediment. Magnification 100.



Figure A.122: Clay and sediment organics in Wilhelmina Polder sediment. Note that sediment organics are all white. Magnification 100.



Figure A.123: Quartz and sediment organics in Zelhem sediment. Magnification 50.



Figure A.124: Quartz and sediment organics in Zelhem sediment. Magnification 50.



Figure A.125: Hyphae on plant remains in Zelhem sediment. Magnification 50.

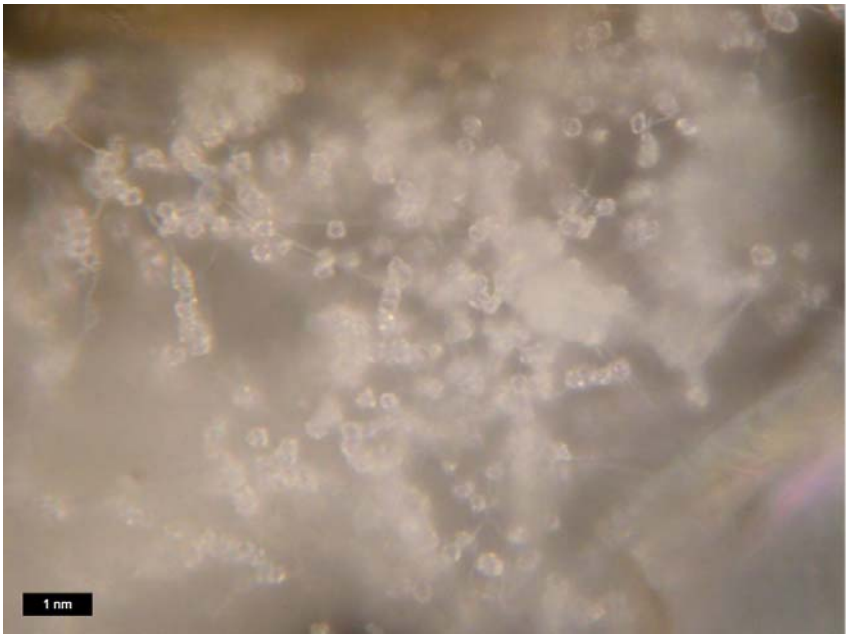


Figure A.126: Spores in Zelhem sediment. These spores can be confused with starch grains. However, these spores are much smaller and do not have a clear extinction cross. Magnification 800, brightfield light.

B APPENDIX: RESULTS FROM THE EXPERIMENTAL SAMPLE

This appendix presents the micrographs from Chapter Four, which are the results from the replicated sample. The appendix consists of micrographs and distribution maps on which the locations of all residues are indicated per stone tool. Unless stated otherwise all micrographs are taken in darkfield light using no filters. All the tools in the Figures are orientated with the distal end facing upwards. The key for the distribution maps is as follows:

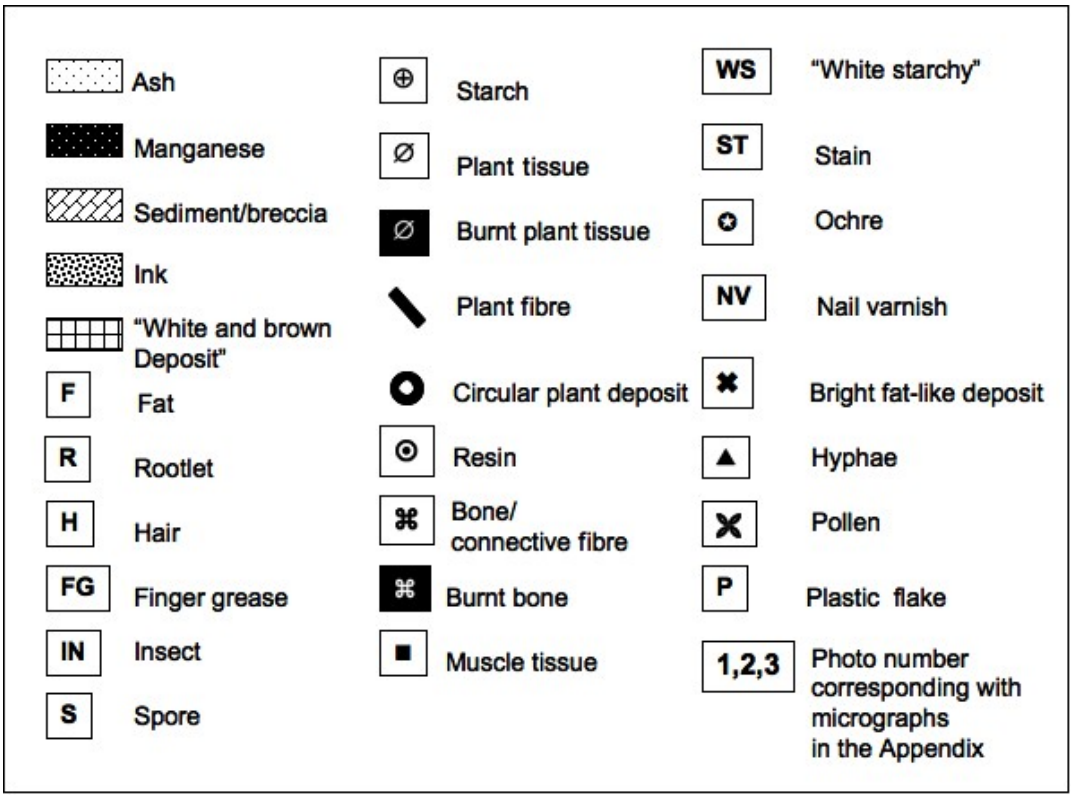


Figure B.1: Key to the distribution maps in Appendix B.

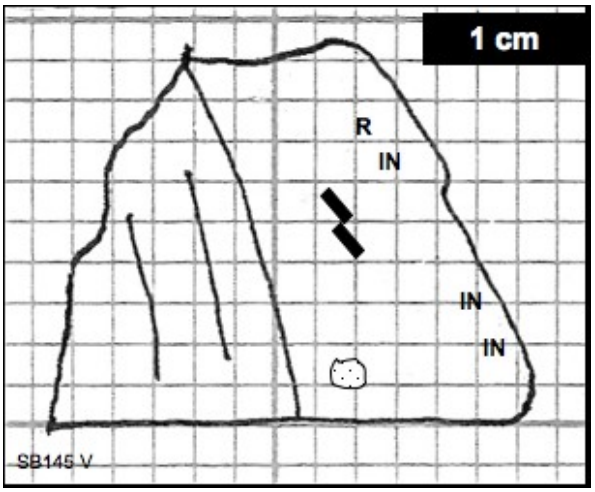


Figure B.2: Distribution map for blank flake SB145, ventral side. For key see Figure B.1.

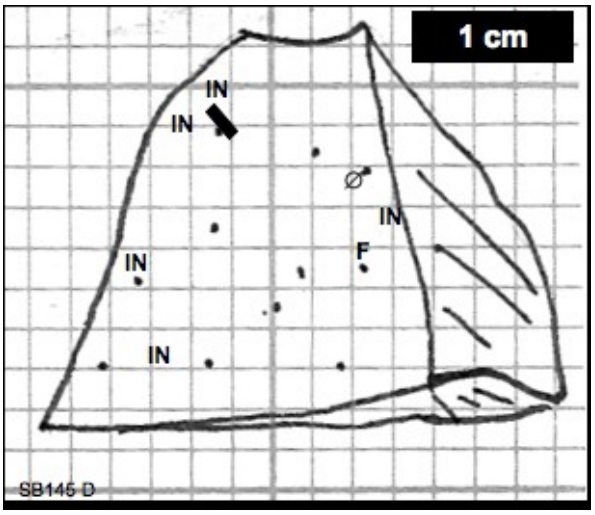


Figure B.3: Distribution map for blank flake SB145, dorsal side. Note the random distribution of insect remains. For key see Figure B.1.

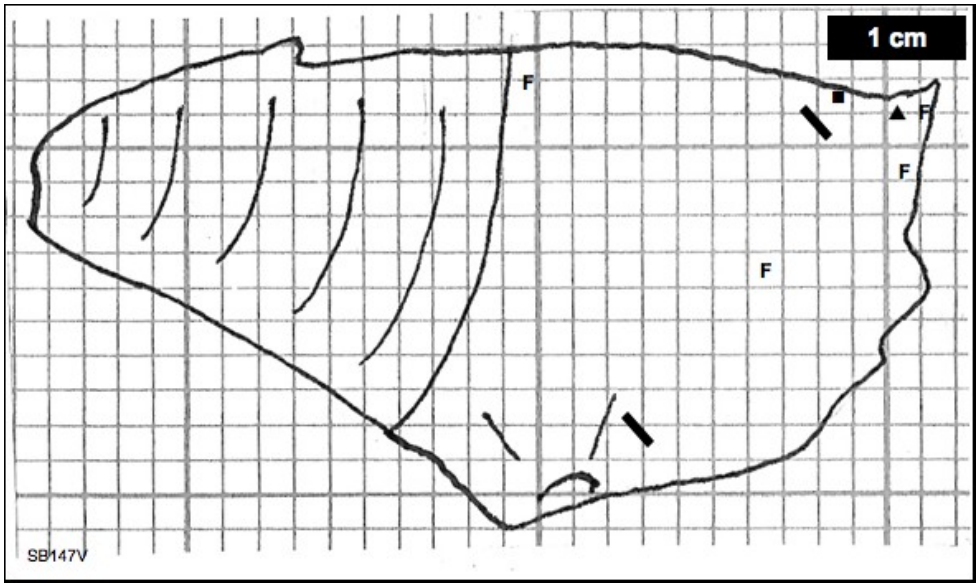


Figure B.4: Distribution map for blank flake SB147, ventral side. For key see Figure B.1.

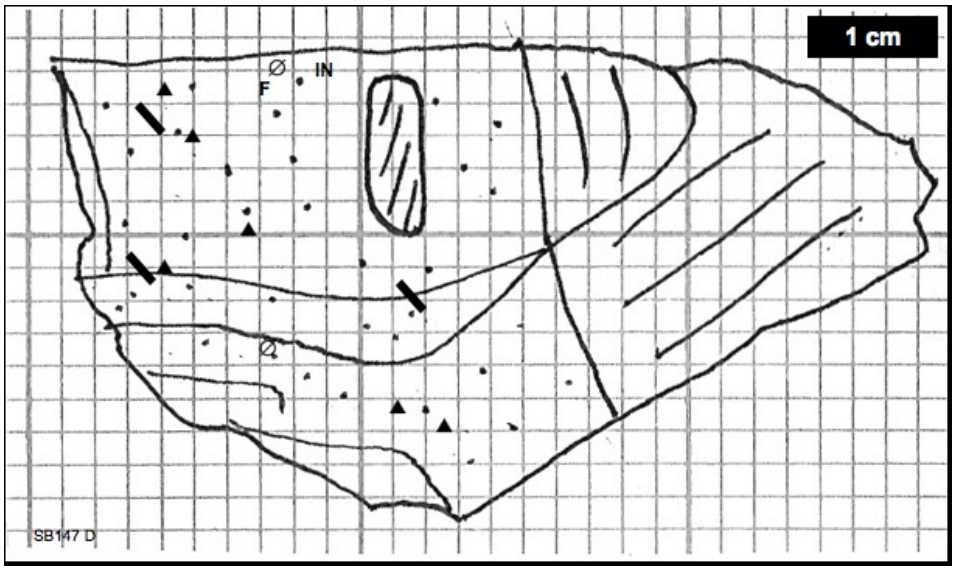


Figure B.5: Distribution map blank flake SB147, dorsal side. The micro-remains on this blank do not appear to have a meaningful distribution and there is no association between, for example, the fat, insect and plant tissue remains at the distal end. For key see Figure B.1.

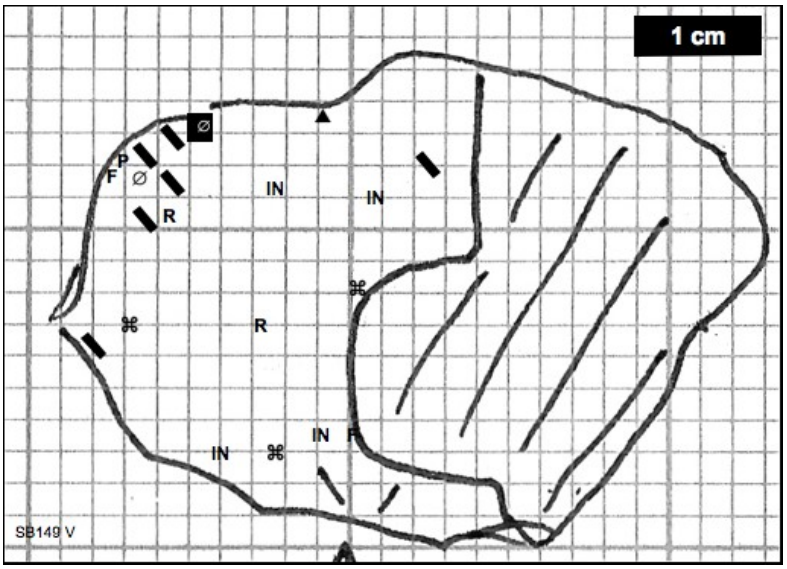


Figure B.6: Distribution map for blank flake SB149, ventral side. There are several plant fibres present along the edge. However, the combination with plastic, charcoal, plant tissue, fat, rootlets, insect remains and bone reveal the contaminant nature of these fibres. For key see Figure B.1.

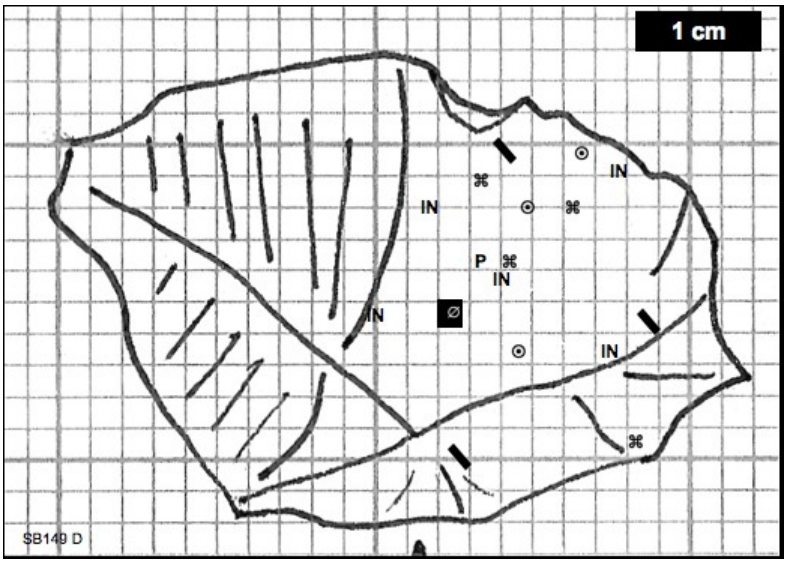


Figure B.7: Distribution map for blank flake SB149, dorsal side. Note that resin or a resin-like deposit can also be a contaminant. For key see Figure B.1.

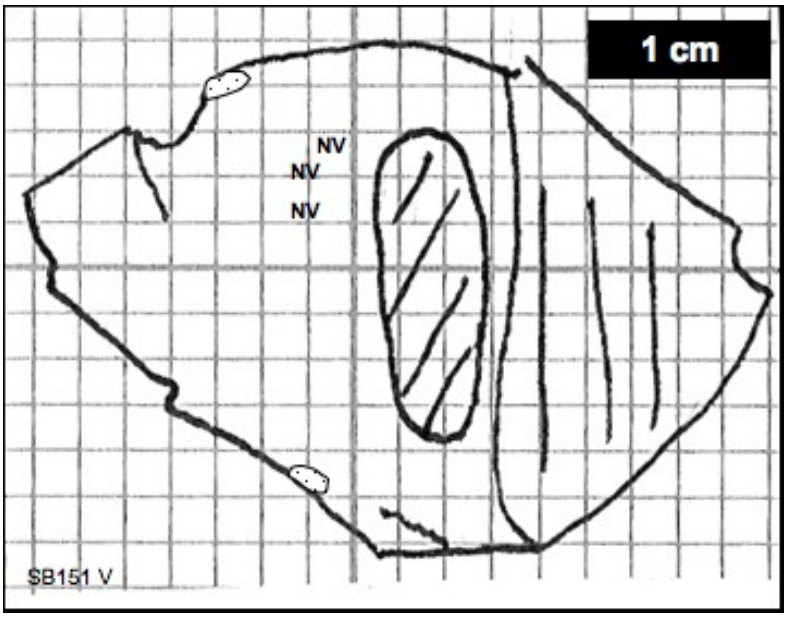


Figure B.8: Distribution map for blank flake SB151, ventral side. For key see Figure B.1.

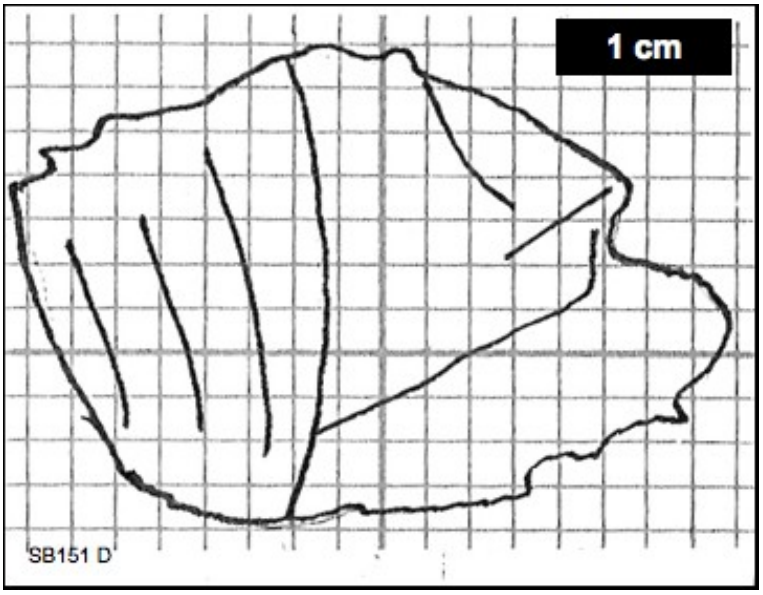


Figure B.9: Distribution map for blank flake SB151, dorsal side. For key see Figure B.1.

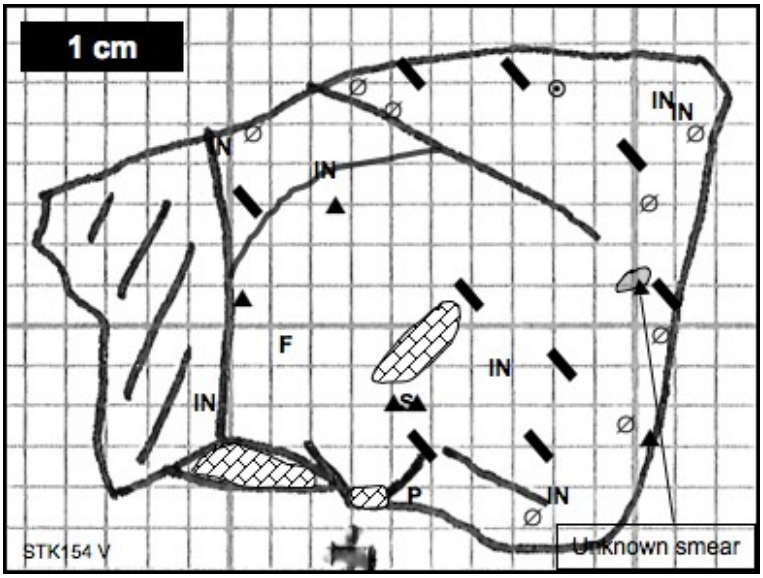


Figure B.10: Distribution map for blank flake STK154, ventral side. Note the random distribution of plant fibres and other remains. For key see Figure B.1.

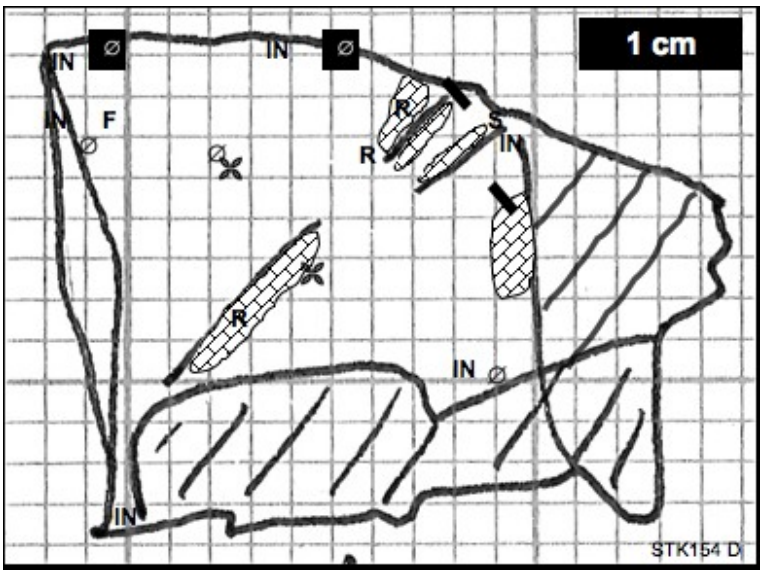


Figure B.11: Distribution map for blank flake STK154, dorsal side. For key see Figure B.1.

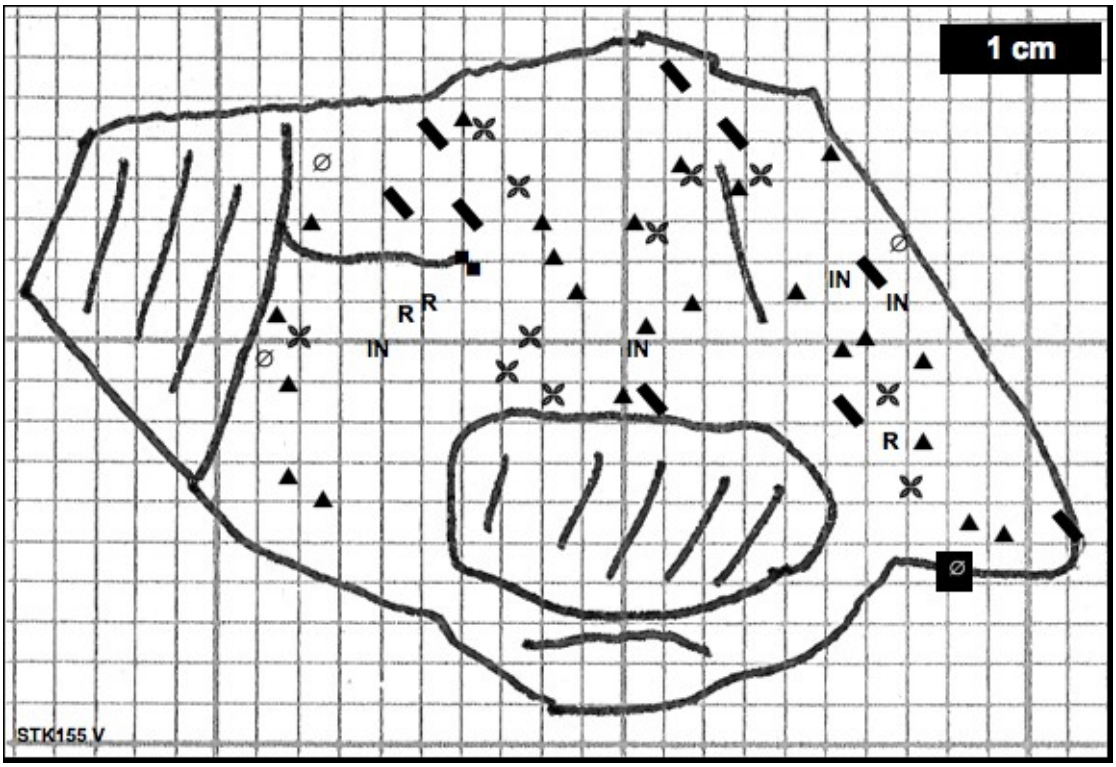


Figure B.12: Distribution map for blank flake STK155, ventral side. The remains on this artefact have a random distribution. In addition, the muscle tissue remains, just left of the middle, are isolated remains. Note that hyphae and pollen are abundant and therefore they should not be used to reconstruct processed materials. For key see Figure B.1.

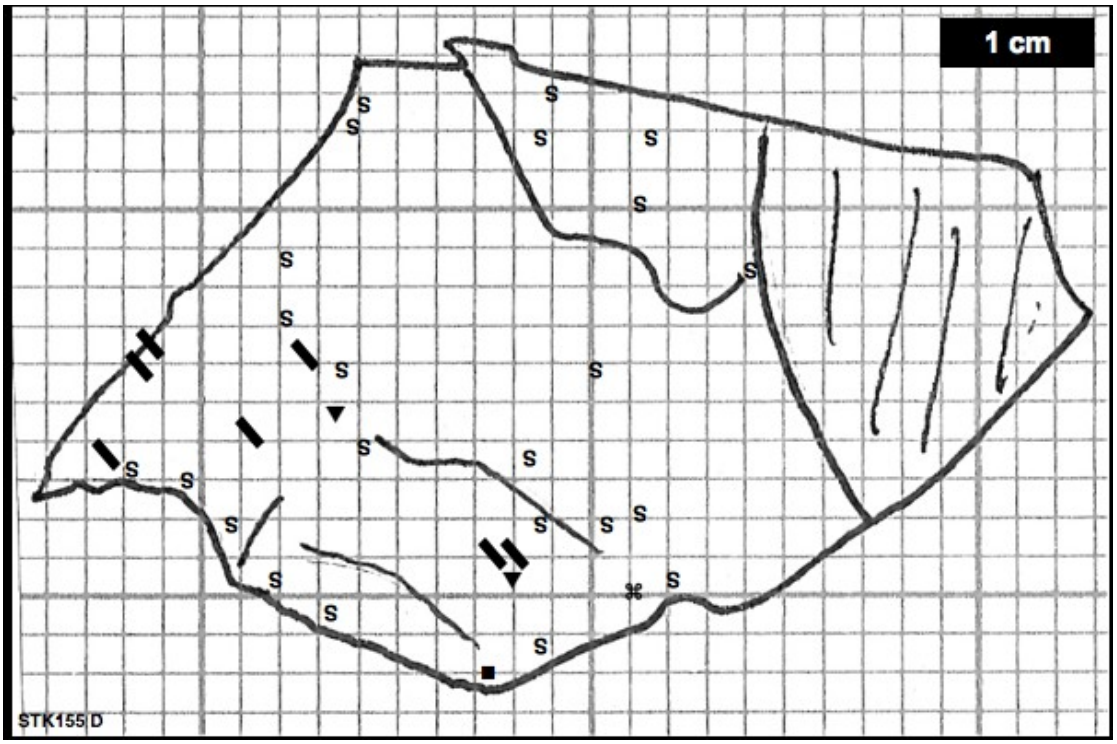


Figure B.13: Distribution map for blank flake STK155, dorsal side. Note how the random distribution and association of contaminants, like spores and hyphae, rule out the isolated deposits of muscle tissue and bone as use-related residues. For key see Figure B.1.

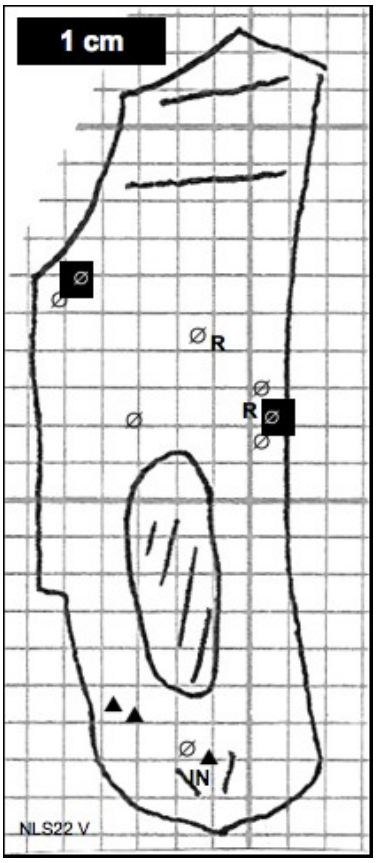


Figure B.14: Distribution map for blank flake NLS22, ventral side. For key see Figure B.1.

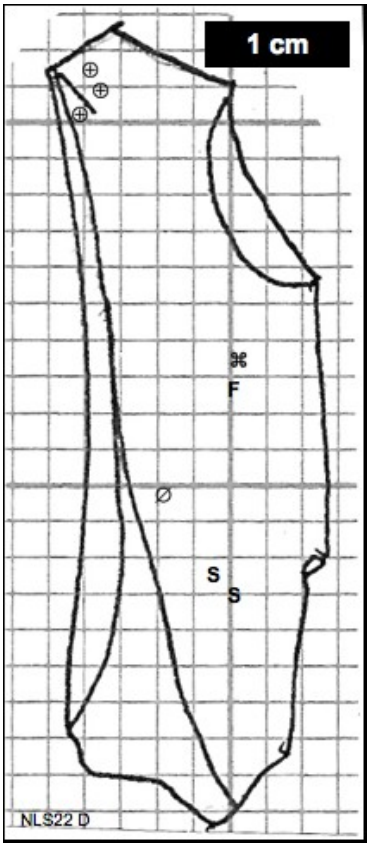


Figure B.15: Distribution map for blank flake NLS22, dorsal side. Note that starch grains can be contaminants. For key see Figure B.1.

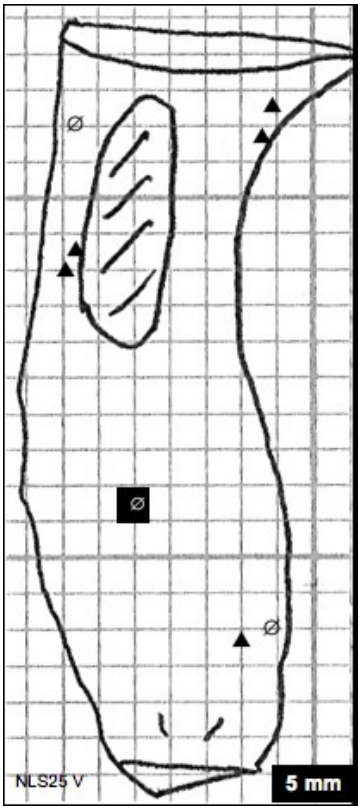


Figure B.16: Distribution map for blank flake NLS25, ventral side. For key see Figure B.1.

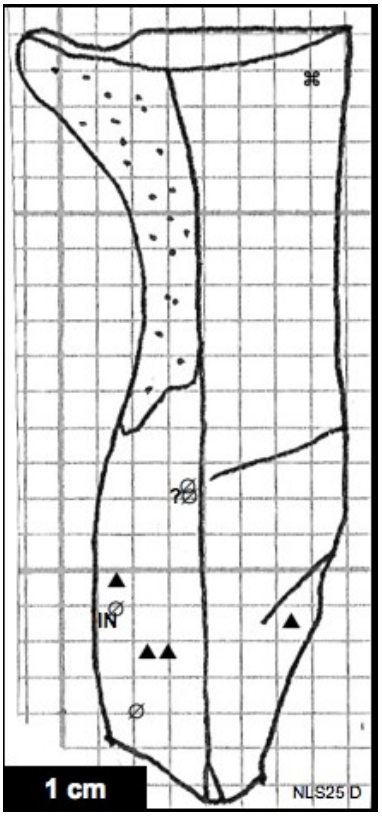


Figure B.17: Distribution map for blank flake NLS25, dorsal side. For key see Figure B.1.

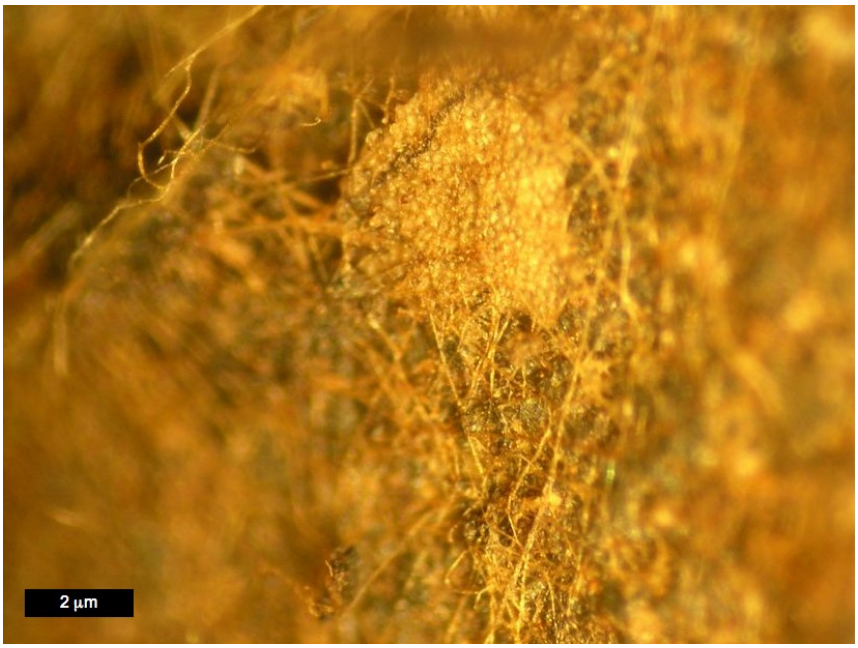


Figure B.18: Micrograph of hyphae on insect remains on a blank. Hyphae grow on sediment remains and use-related residues and therefore cannot be used to reconstruct processed materials. Magnification 50.

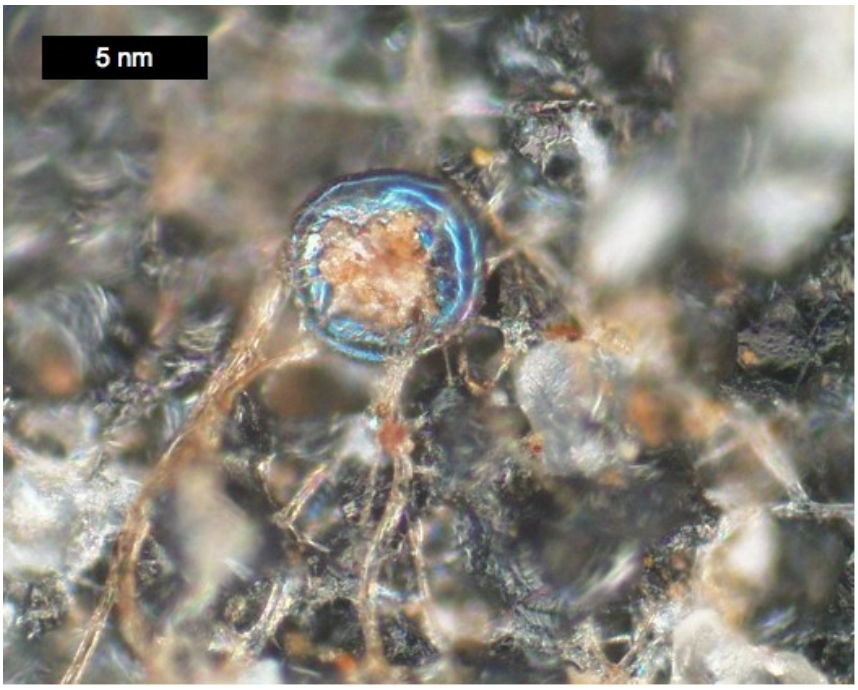


Figure B.19: Micrograph of hyphae growing on pollen on a blank flake. Hyphae grow on sediment remains and use-related residues and therefore cannot be used to reconstruct processed materials. Magnification 200, brightfield cross-polarised light.

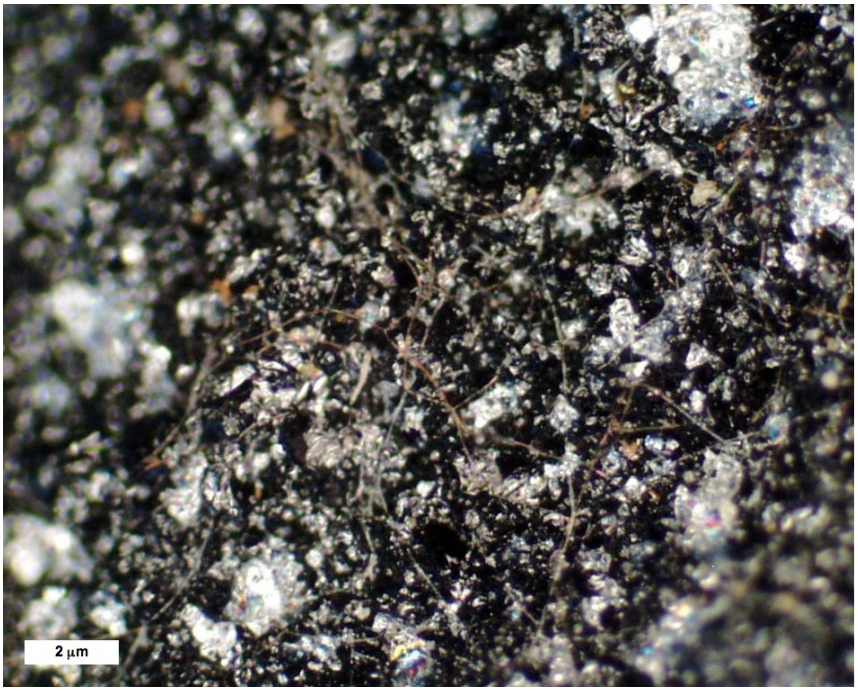


Figure B.20: Micrograph of hyphae on Sterkfontein blank STK155. Hyphae are ever present in the soil and on stone artefacts. Magnification 50

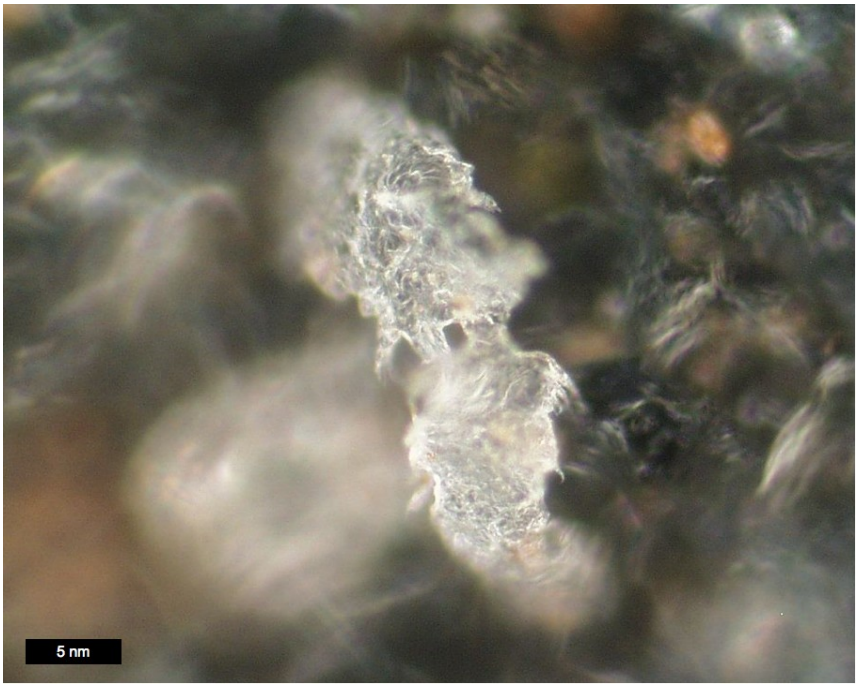


Figure B.21: Micrograph fat droplets on Sterkfontein blank STK154. Animal type residues are usually not regarded as contaminants, but this find demonstrates that animal remains could also be contamination. Magnification 200.

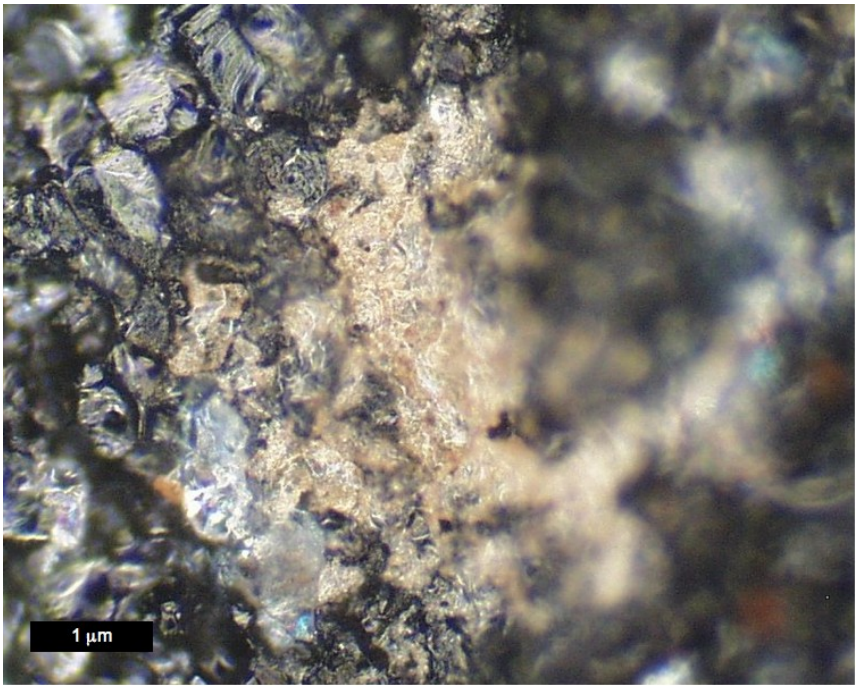


Figure B.22: Micrograph of a muscle tissue-like deposit on Sterkfontein blank STK155. Animal type residues are usually not regarded as contaminants, but this find demonstrates that animal remains could also be contamination or part of a misidentification. Magnification 100.

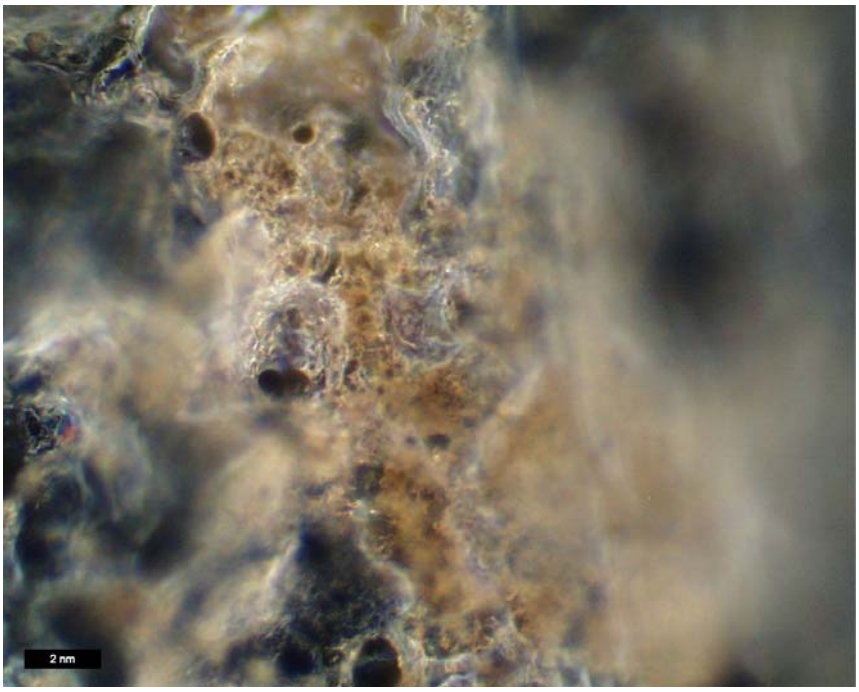


Figure B.23: Micrograph of a muscle tissue-like deposit on Sterkfontein blank STK155. Magnification 500, brightfield polarised light.

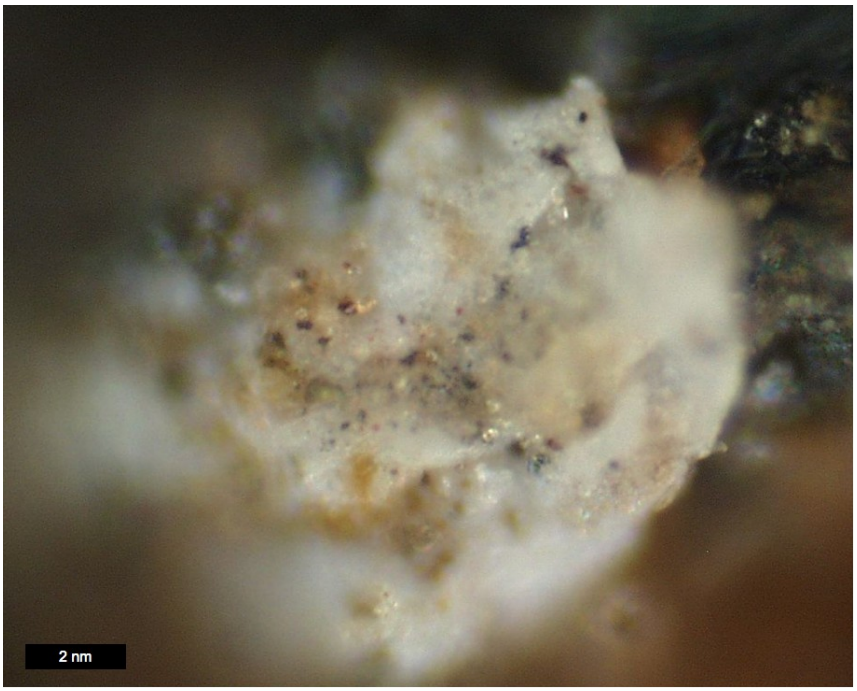


Figure B.24: Micrograph of a fat-like deposit on Sibudu blank SB149. Fat-like deposits are present on both the Sterkfontein and Sibudu blanks and these finds caution against an instant conclusion that all animal type residues are use-related. Magnification 500.



Figure B.25: Micrograph of a fibre similar to a connective fibre on Sibudu blank SB149. Not only muscle tissue and fat, but also bone and connective fibres can be animal type contaminants. Magnification 200.

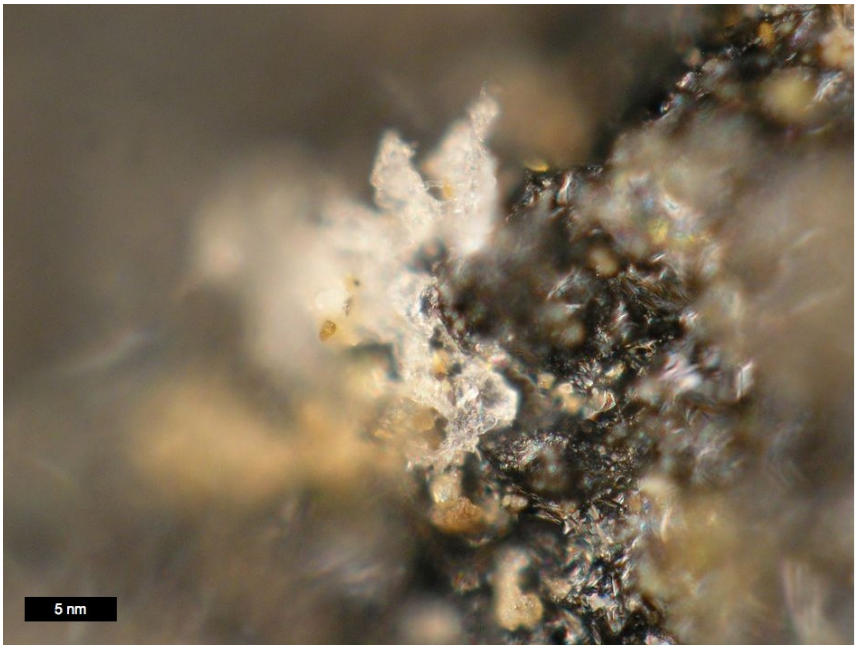


Figure B.26: Micrograph of a fatty bone-like deposit on Sibudu blank SB156. Not only muscle tissue and fat, but also bone and connective fibres can be animal type contaminants. Magnification 200.

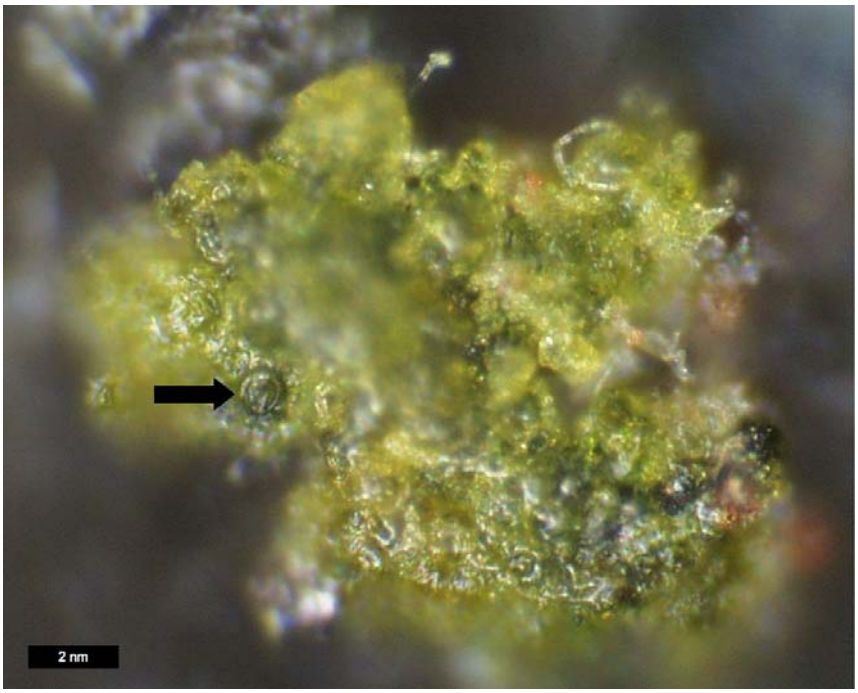


Figure B.27: Micrograph a green fat-like deposit with pollen (indicated with arrow) on Sterkfontein blank SB154. Magnification 500.

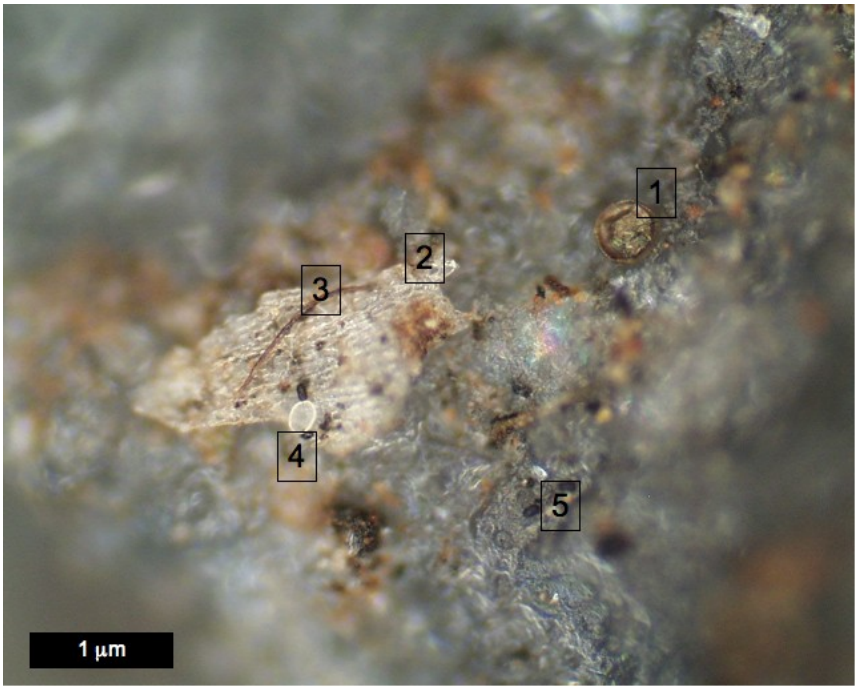


Figure B.28: Micrograph of sediment organics Sterkfontein blank STK154. Note that plant tissue (2) is surrounded by obvious contaminants such as pollen (1 and 4), hyphae (3) and spores (5); indicating that the plant tissue could originate from the sediment too. Magnification 50.

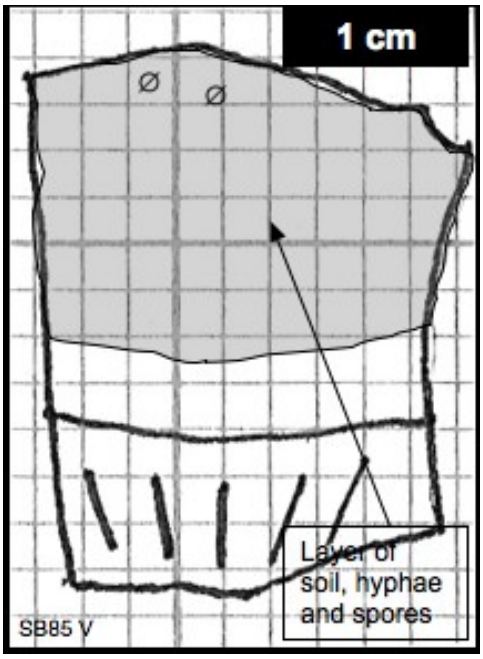


Figure B.29: Distribution map for SB85, ventral side. Note the thick layer of hyphae and spores, which obscures the starch grains. For key see Figure B.1.

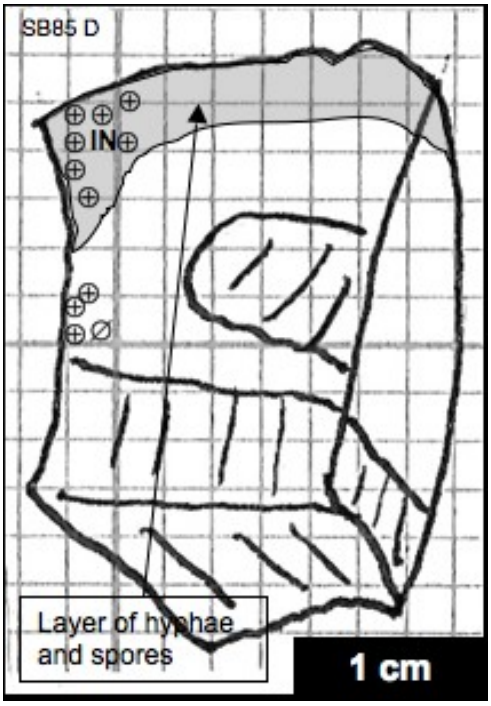


Figure B.30: Distribution map for SB85, dorsal side. Note the thick layer of hyphae and spores, which obscures the starch grains. For key see Figure B.1.

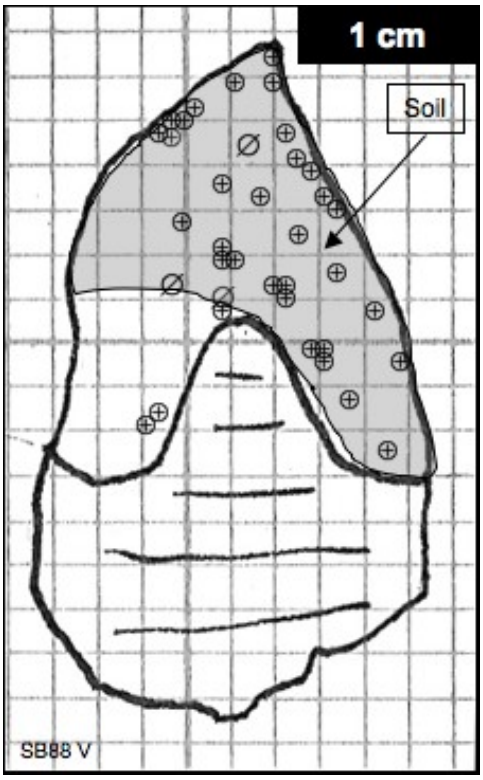


Figure B.31: Distribution map for SB88, ventral side. Thick sediment layers attached to the moist residue layer. For key see Figure B.1.

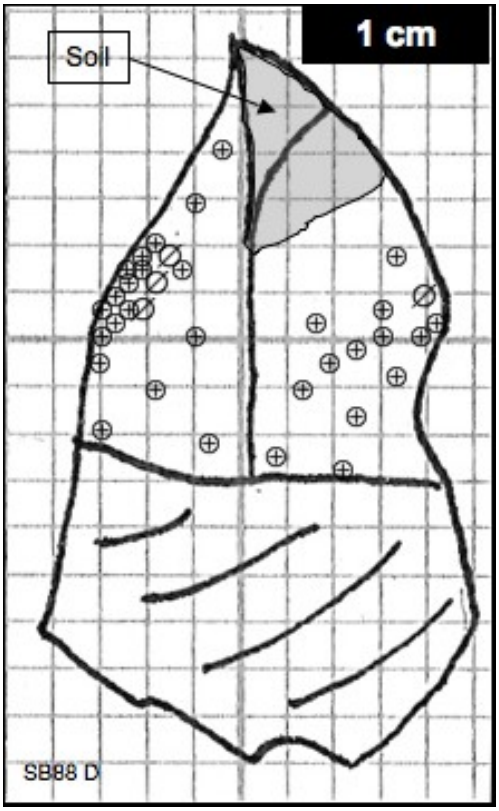


Figure B.32: Distribution map for SB88, dorsal side. For key see Figure B.1.

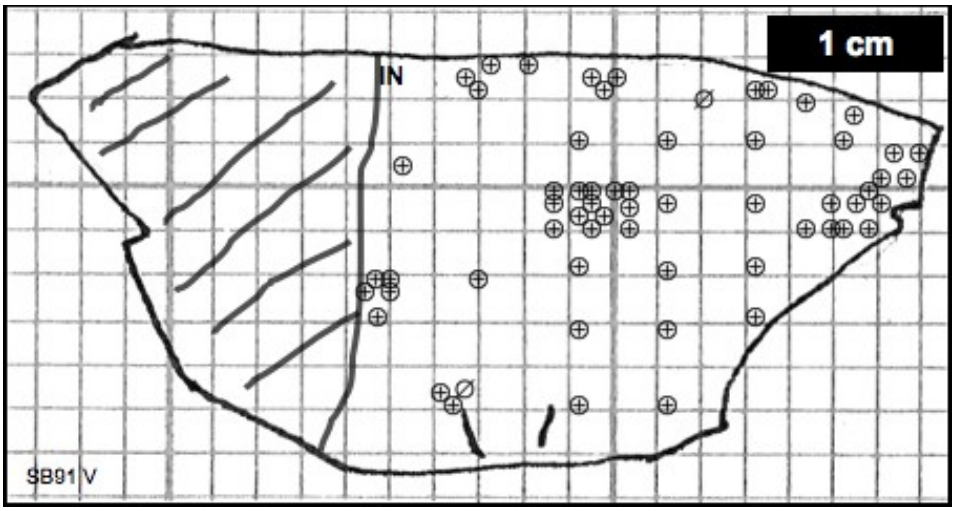


Figure B.33: Distribution map for SB91, ventral side. For key see Figure B.1.

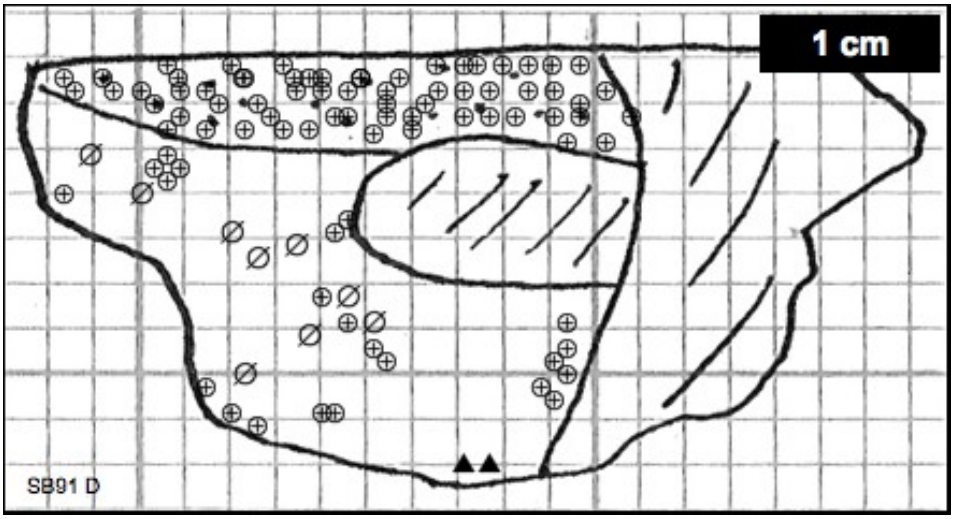


Figure B.34: Distribution map for SB91, dorsal side. Although use-related residues are generally concentrated along the working edge, starch type residues are present all over the tool's surface. For key see Figure B.1.

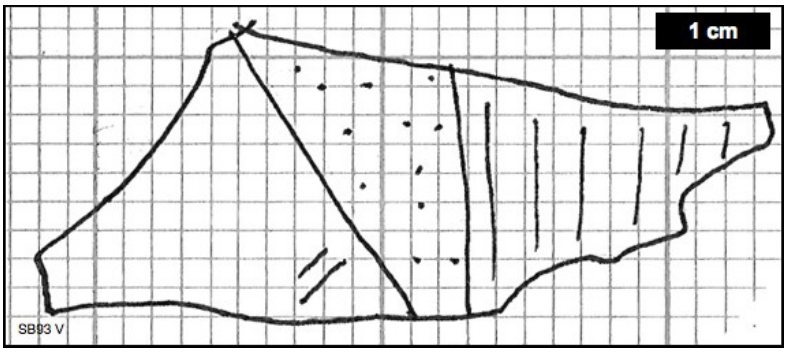


Figure B.35: Distribution map for SB93, ventral side. No residues are present on the ventral side. For key see Figure B.1.

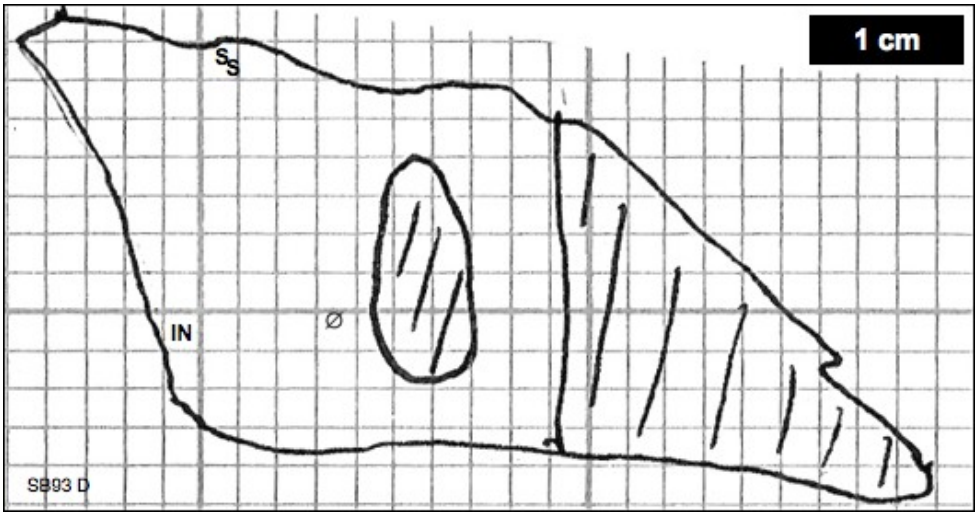


Figure B.36: Distribution map for SB93, dorsal side. For key see Figure B.1.

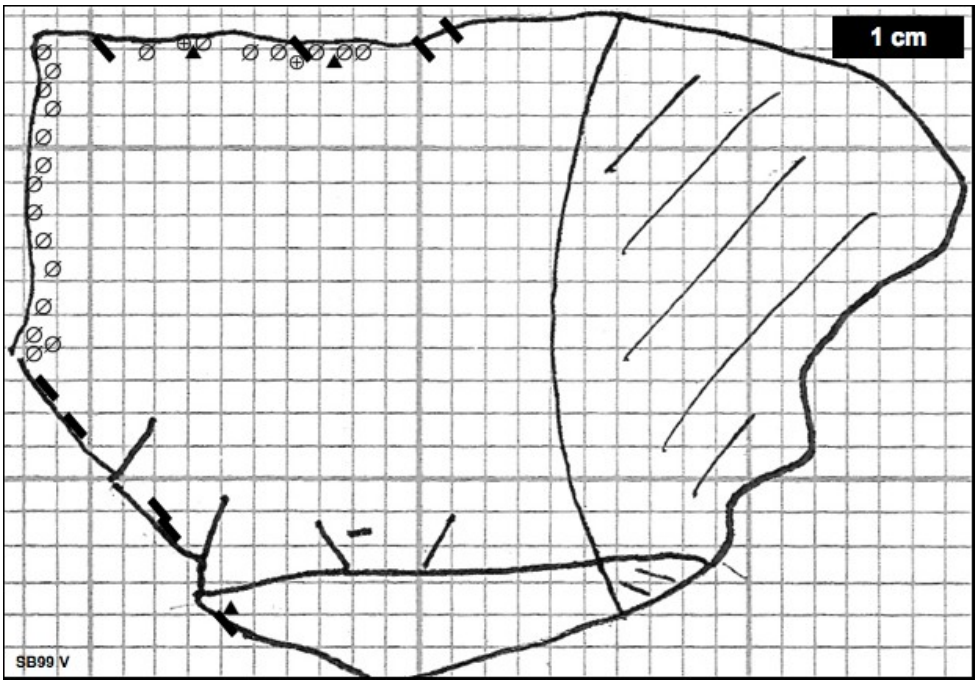


Figure B.37: Distribution map for SB99, ventral side. Note that the use-related starch and plant fibres are confined to the edges. For key see Figure B.1.

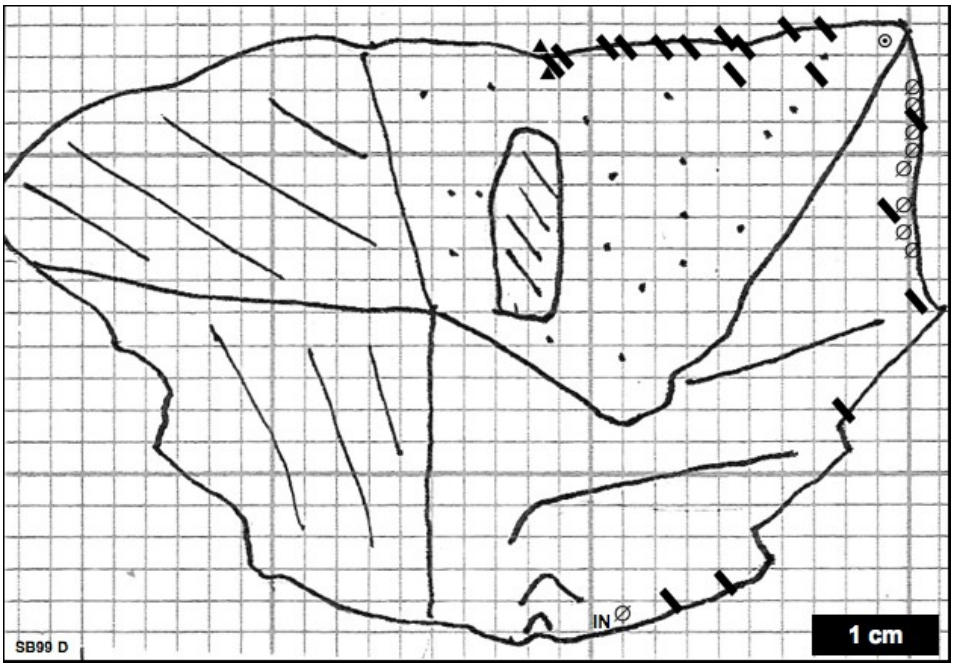


Figure B.38: Distribution map for SB99, dorsal side. Note that the use-related starch and plant fibres are confined to the edges. For key see Figure B.1.

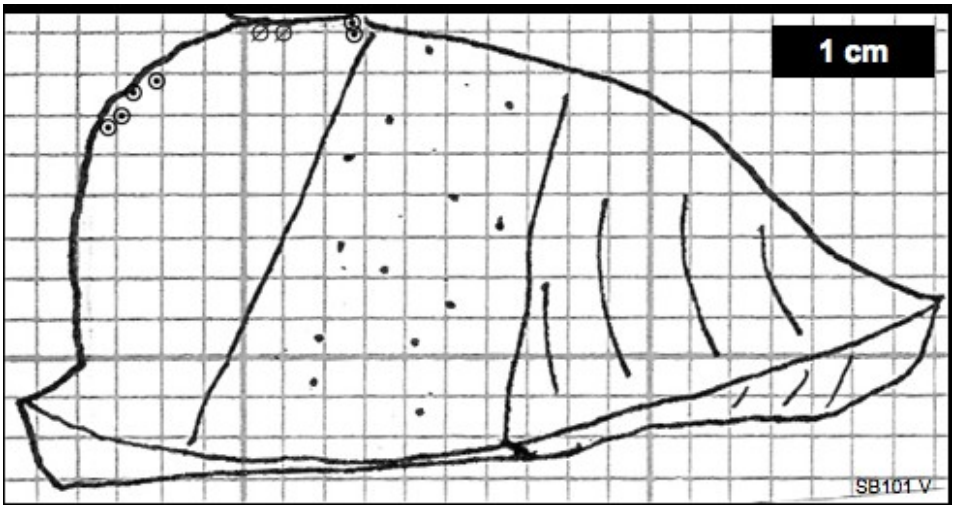


Figure B.39: Distribution map for SB101, ventral side. For key see Figure B.1.

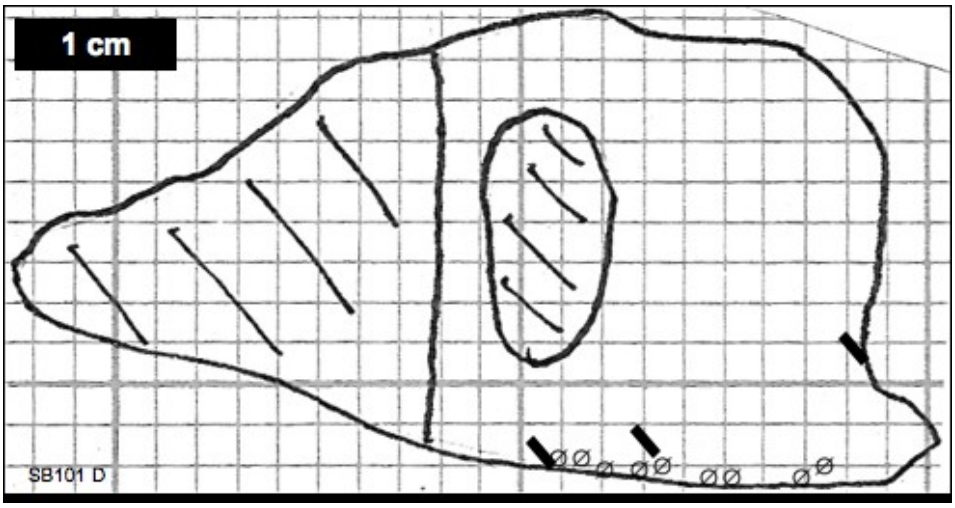


Figure B.40: Distribution map for SB101, dorsal side. Note that the use-related plant tissue residues are confined to the edges. For key see Figure B.1.

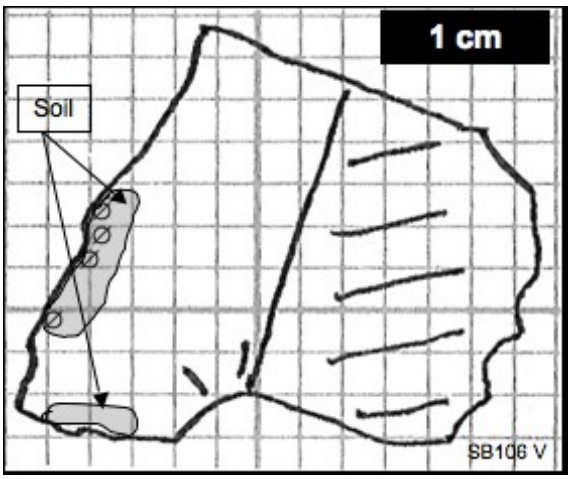


Figure B.41: Distribution map for SB106, ventral side. Again sediment is attached to the use-related residues. For key see Figure B.1.

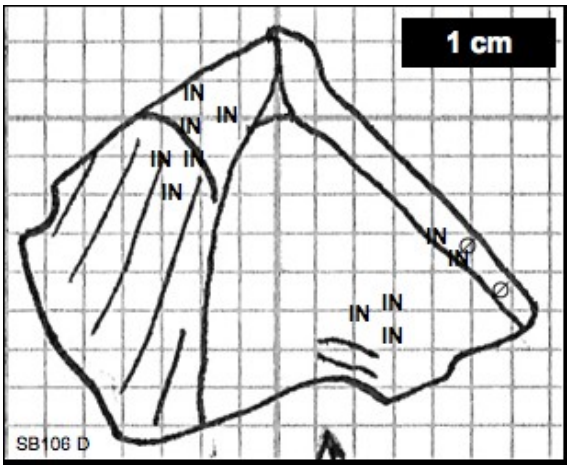


Figure B.42: Distribution map for SB106, dorsal side. Many insect remains are present on this tool. For key see Figure B.1.

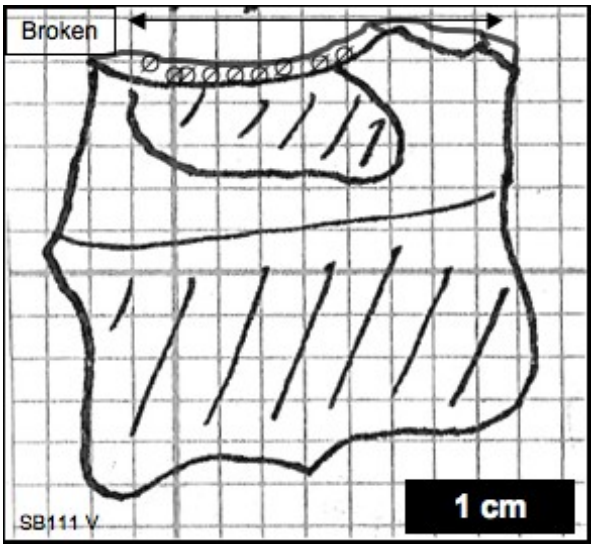


Figure B.43: Distribution map for SB111, ventral side. Note that the use-related starch residues are confined to the edges. For key see Figure B.1.

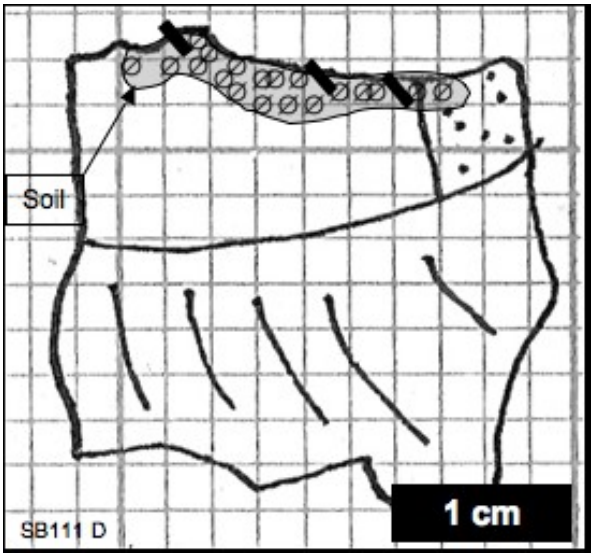


Figure B.44: Distribution map for SB111, dorsal side. Note that the use-related starch residues are confined to the edges. Sediment covers the starch type residues. For key see Figure B.1.

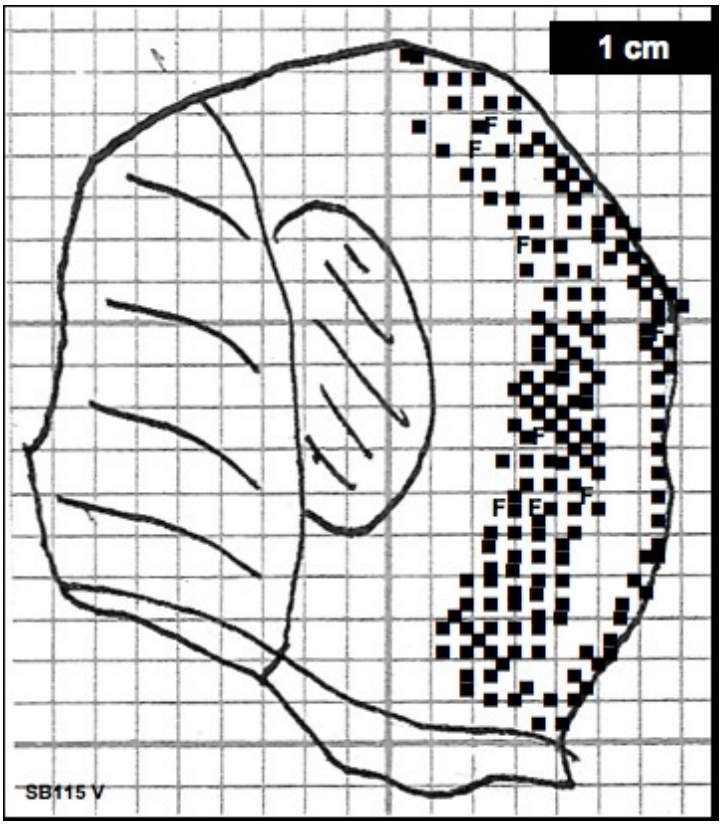


Figure B.45: Distribution map for SB115, ventral side. Note that the use-related muscle tissue residues are confined to the edges. After four weeks, the preservation inside the shelter is extremely well. For key see Figure B.1.

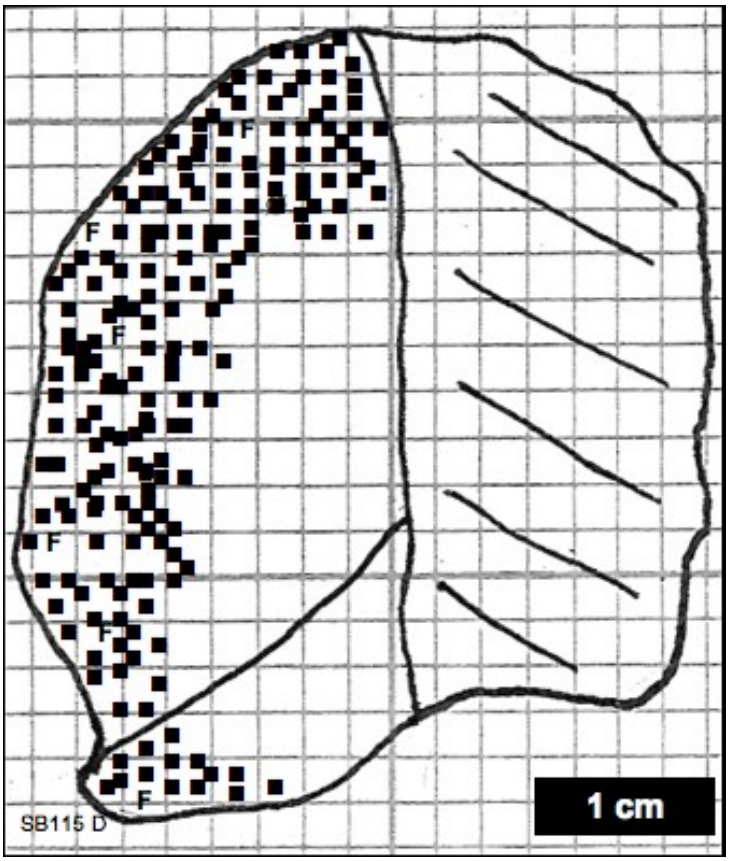


Figure B.46: Distribution map for SB115, dorsal side. Note that the use-related muscle tissue residues are confined to the edges. After four weeks, the preservation inside the shelter is extremely well. For key see Figure B.1.

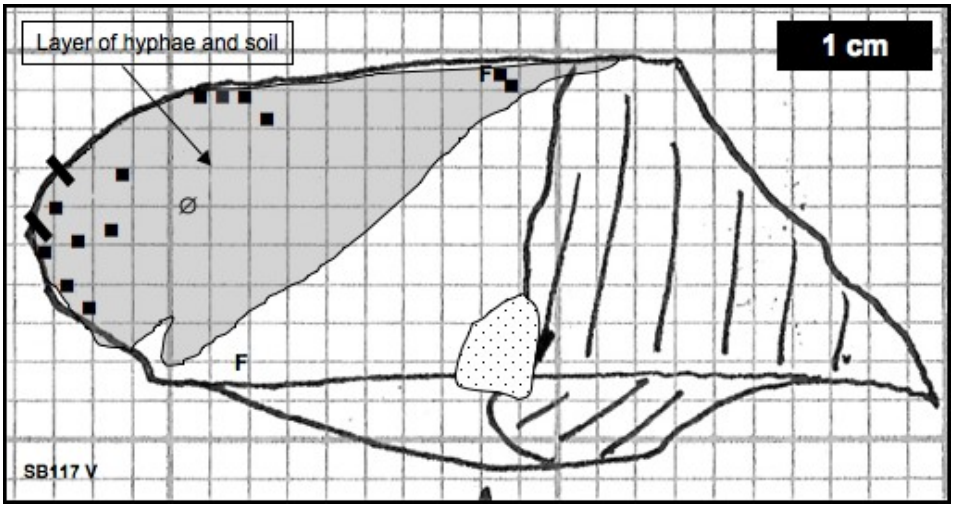


Figure B.47: Distribution map for SB117, ventral side. Note that the use-related muscle tissue residues are confined to the edges. The layer of hyphae and sediment obscures the analysis of the residues. For key see Figure B.1.

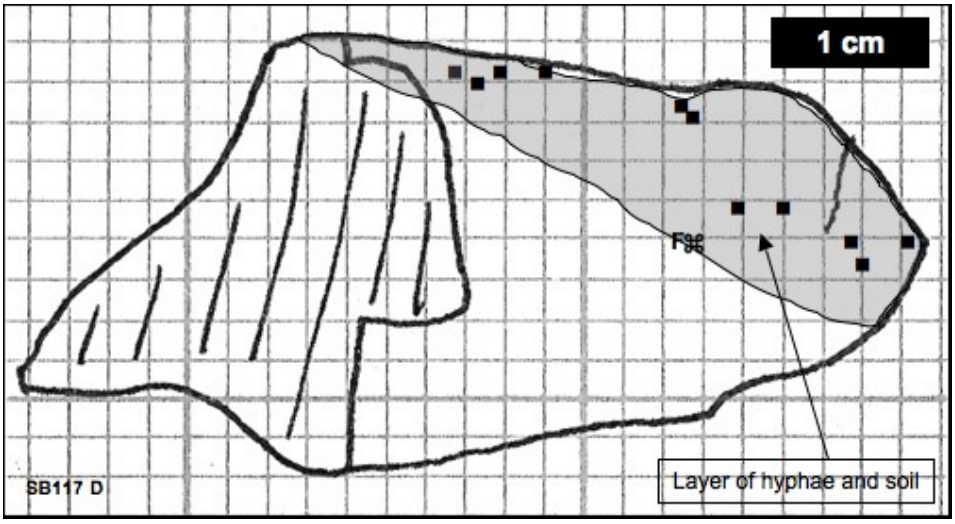


Figure B.48: Distribution map for SB117, dorsal side. Note that the use-related muscle tissue residues are confined to the edges. The layer of hyphae and sediment obscures the analysis of the residues. For key see Figure B.1.

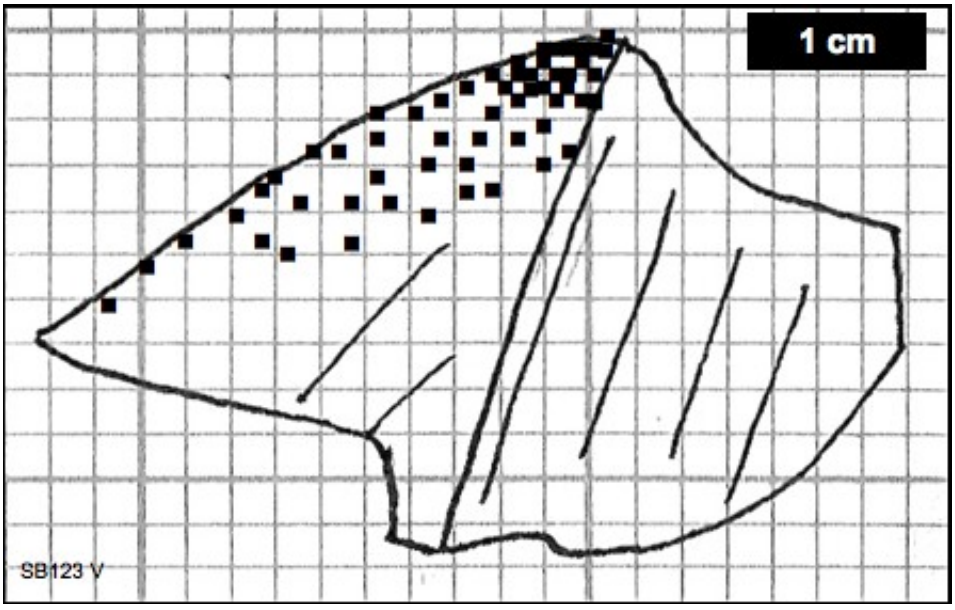


Figure B.49: Distribution map for SB123, ventral side. Note that the use-related muscle tissue residues are confined to the edges. After four weeks, considerably less residues are preserved outside the shelter that inside. For key see Figure B.1.

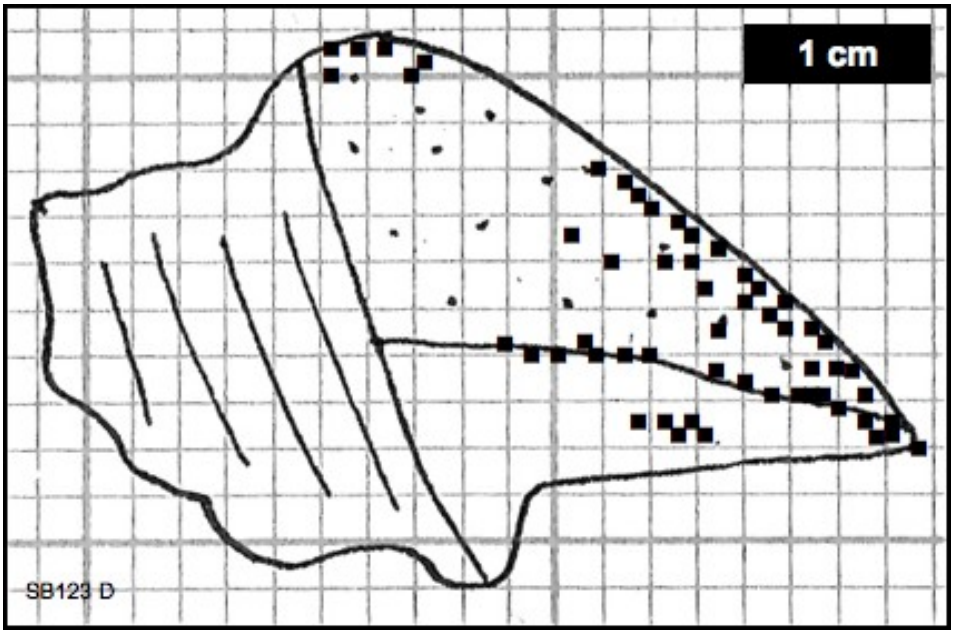


Figure B.50: Distribution map for SB123, dorsal side. For key see Figure B.1.

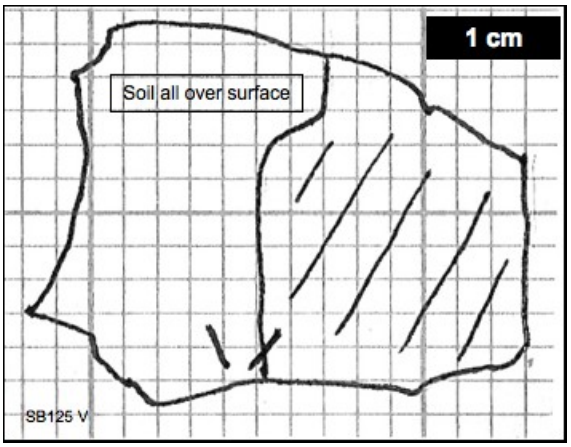


Figure B.51: Distribution map for SB125, ventral side. After one year almost no residues are preserved on tools, which were deposited outside the shelter. For key see Figure B.1.

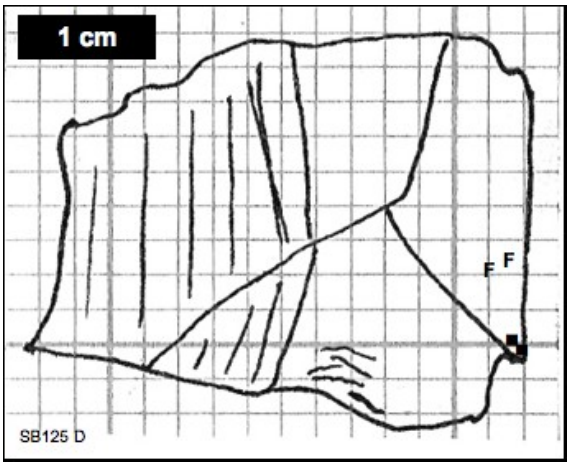


Figure B.52: Distribution map for SB125, dorsal side. After one year almost no residues are preserved on tools, which were deposited outside the shelter. For key see Figure B.1.

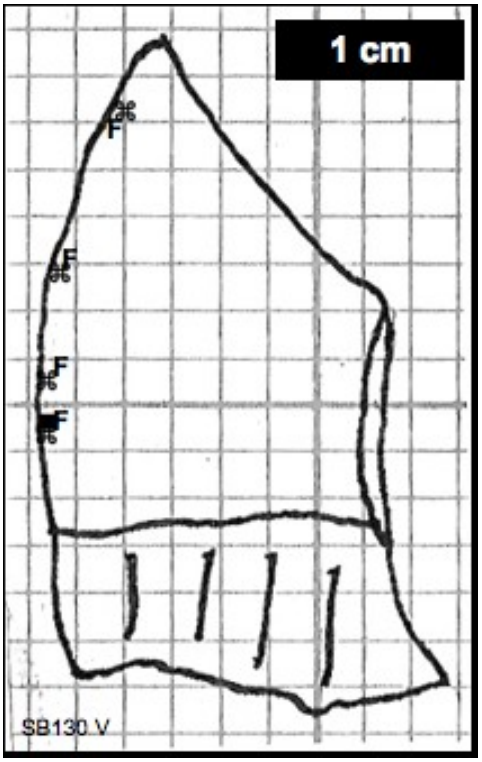


Figure B.53: Distribution map for SB130, ventral side. Note that the use-related residues are confined to the edge. Bone working leaves fewer residues behind on the tool's surface that, for example, processing starchy tubers. For key see Figure B.1.

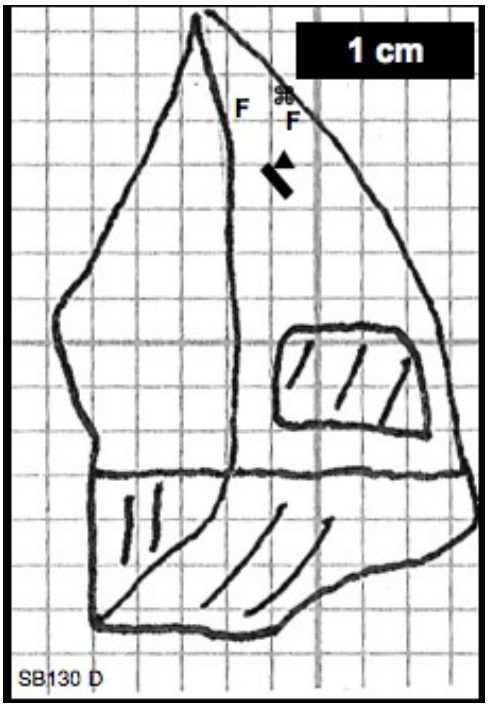


Figure B.54: Distribution map for SB130, dorsal side. The bone and fat residues are confined to the edge. This distribution is typical for scraping bone. For key see Figure B.1.

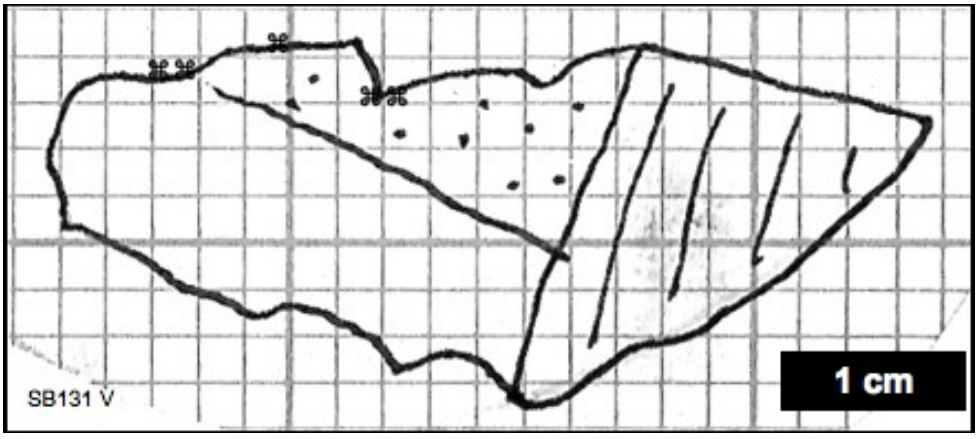


Figure B.55: Distribution map for SB131, ventral side. Note that use-related the bone and fat residues are confined to the edge. For key see Figure B.1.

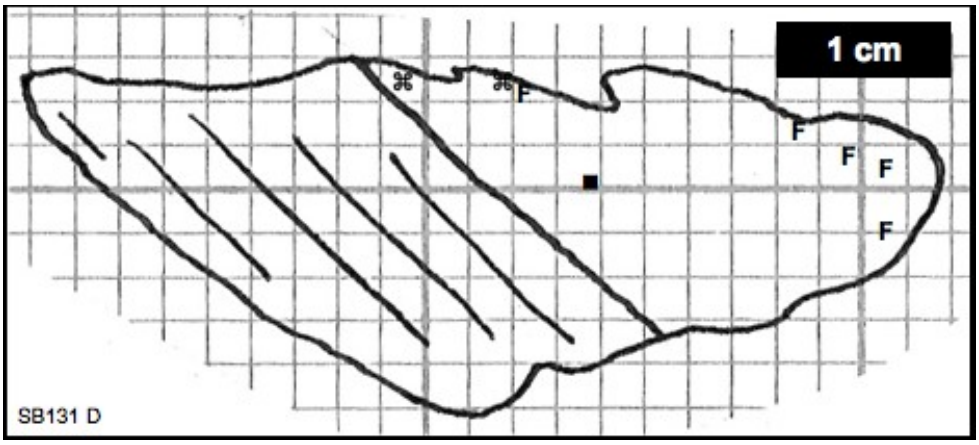


Figure B.56: Distribution map for SB131, dorsal side. Note that the use-related bone and fat residues are confined to the edge. In the middle of the tool is a patch is muscle tissue present. This clearly demonstrates that residues are present in suites. For key see Figure B.1.

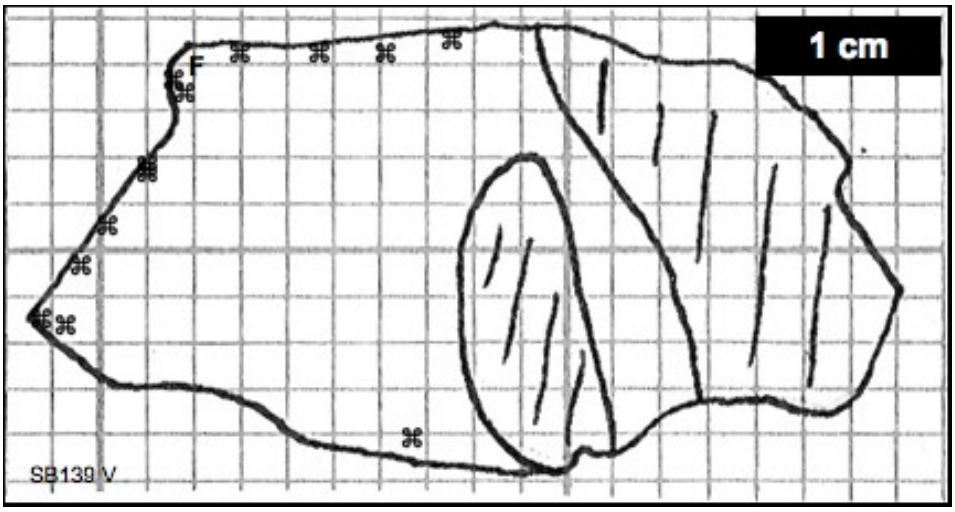


Figure B.57: Distribution map for SB139, ventral side. Note that the use-related bone and fat residues are confined to the edge. For key see Figure B.1.

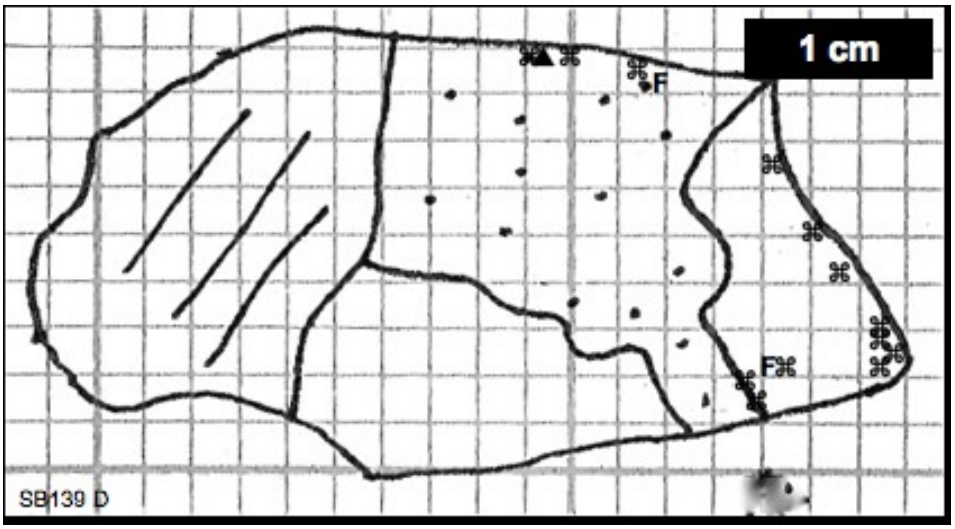


Figure B.58: Distribution map for SB139, dorsal side. Note that the use-related bone and fat residues are confined to the edge. For key see Figure B.1.

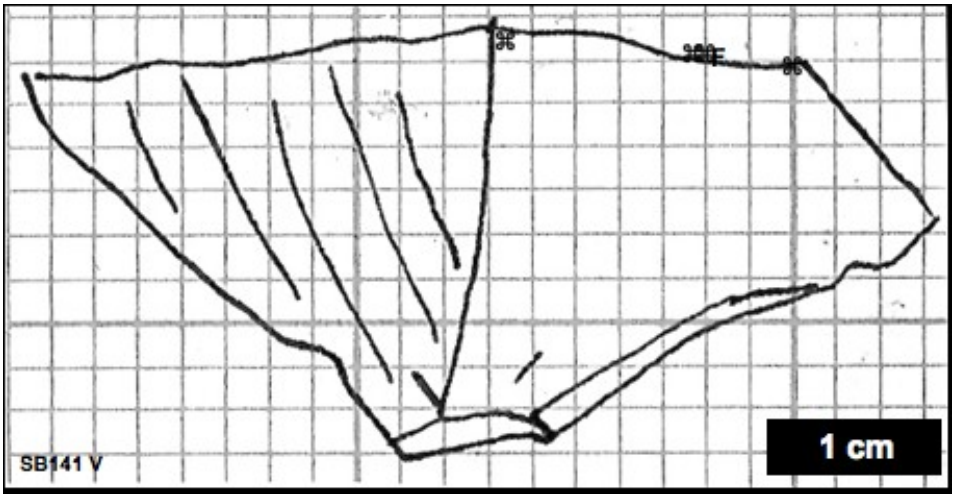


Figure B.59: Distribution map for SB141, ventral side. After one year, few bone and fat residues are preserved outside the shelter. For key see Figure B.1.

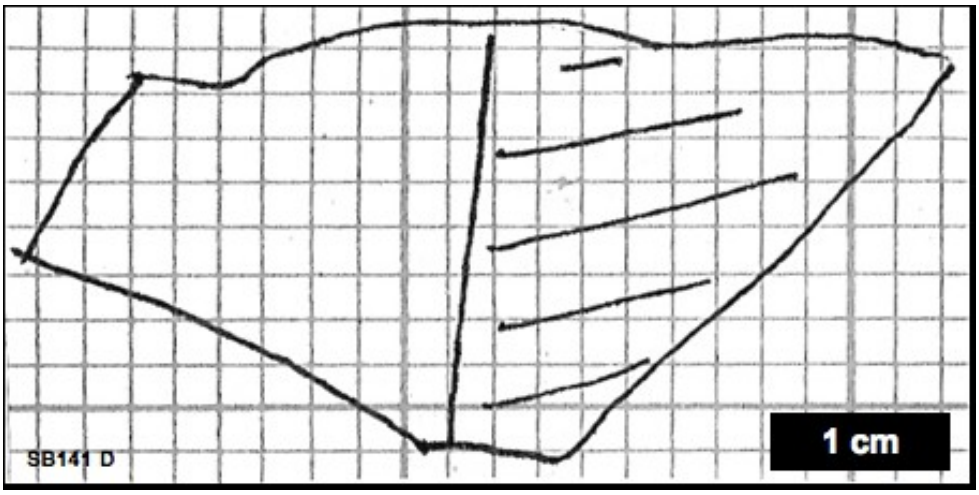


Figure B.60: Distribution map for SB141, dorsal side. After one year, no bone and fat residues are preserved on the dorsal side. For key see Figure B.1.

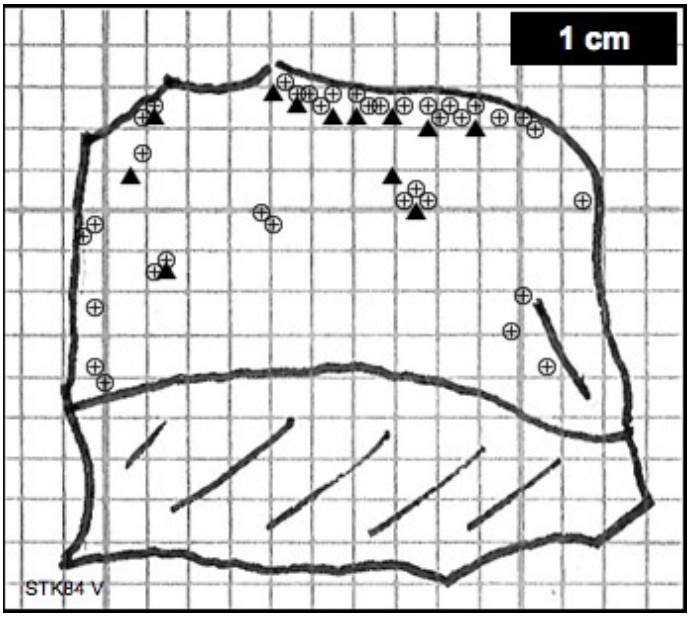


Figure B.61: Distribution map for STK84, ventral side. The use-related starch residues are mainly present along the edges. After four weeks many starch residues are preserved inside the cave. For key see Figure B.1.

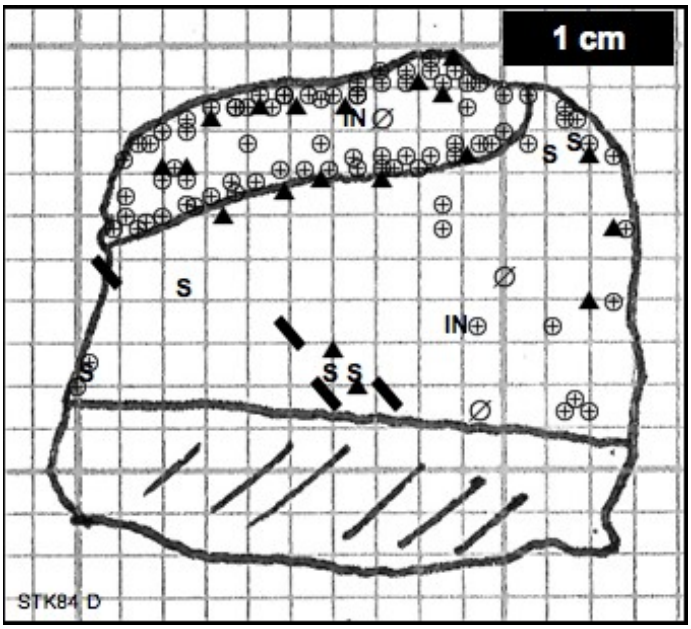


Figure B.62: Distribution map for STK84, dorsal side. After four weeks many starch residues are preserved inside the cave. Use-related residues are typically abundant along the working edge, but starch type residues are more spread-out over the entire tool's surface. For key see Figure B.1.

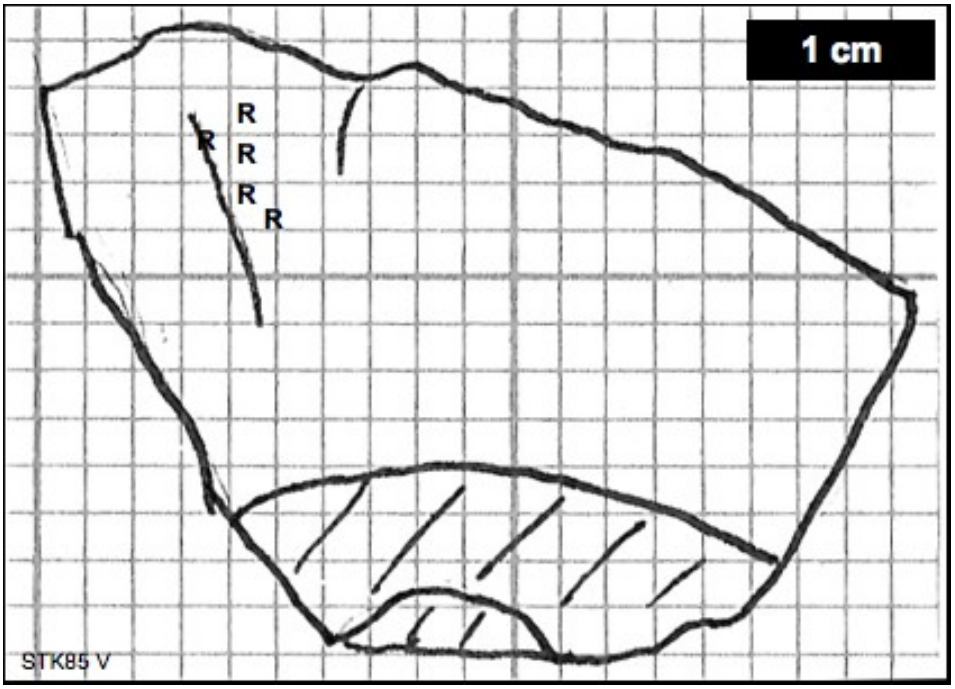


Figure B.63: Distribution map for STK85, ventral side. After one year few starch residues are preserved inside the cave. For key see Figure B.1.

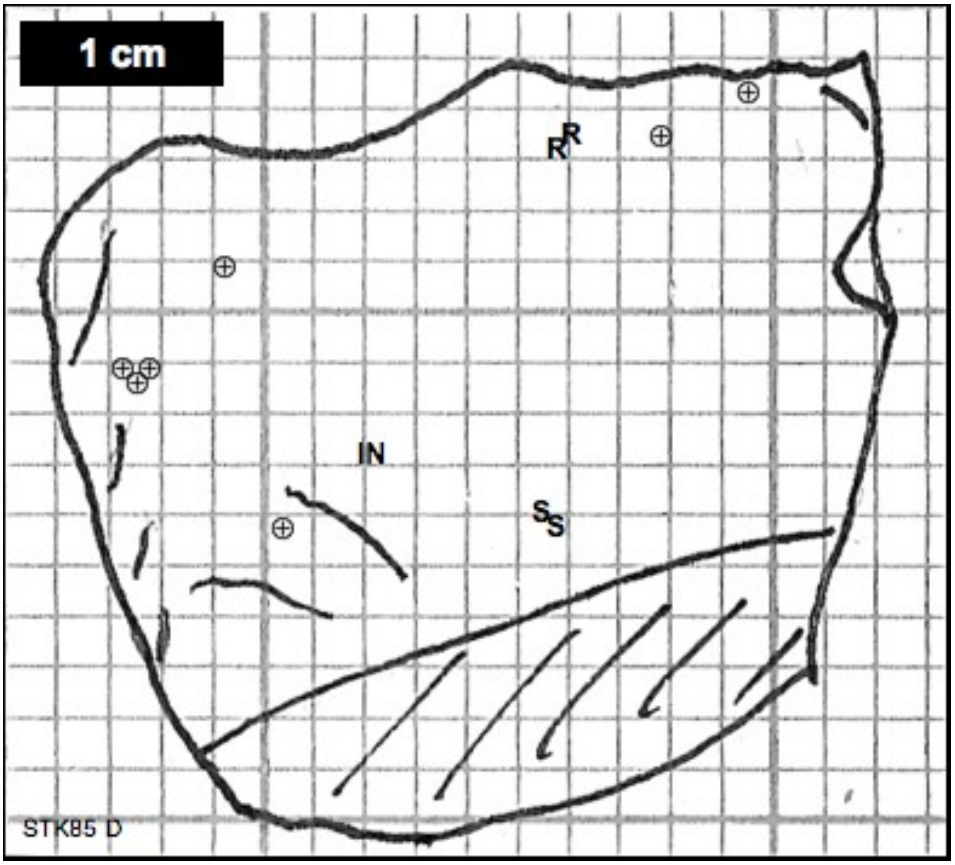


Figure B.64: Distribution map for STK85, dorsal side. After one year few starch residues are preserved inside the cave. For key see Figure B.1.

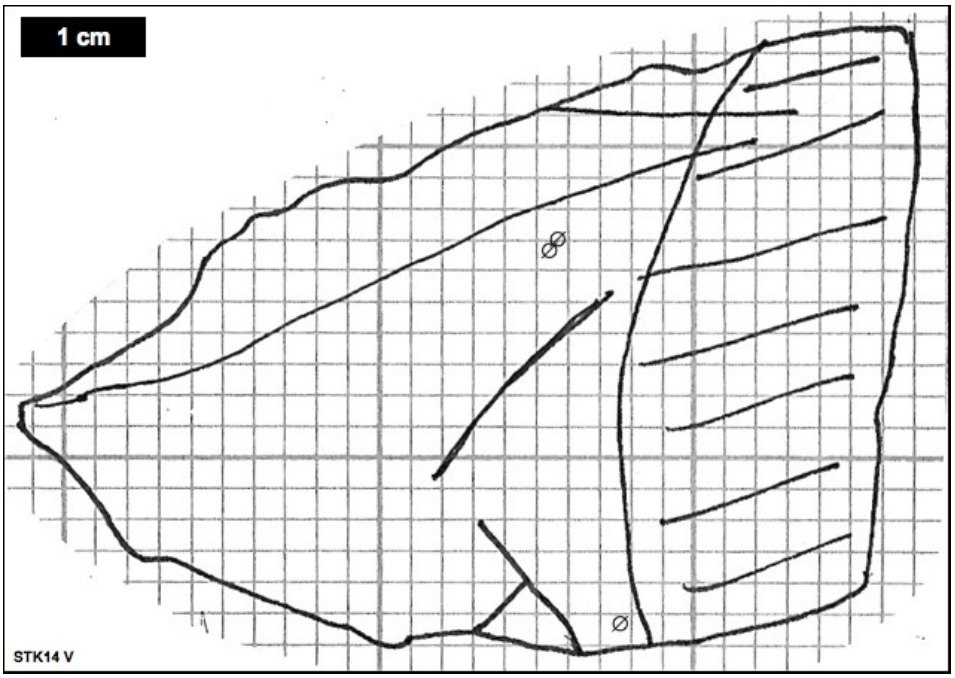


Figure B.65: Distribution map for STK14, ventral side. After one year no starch residues are preserved outside the cave. Note that use-related plant tissue deposits are still present. For key see Figure B.1.

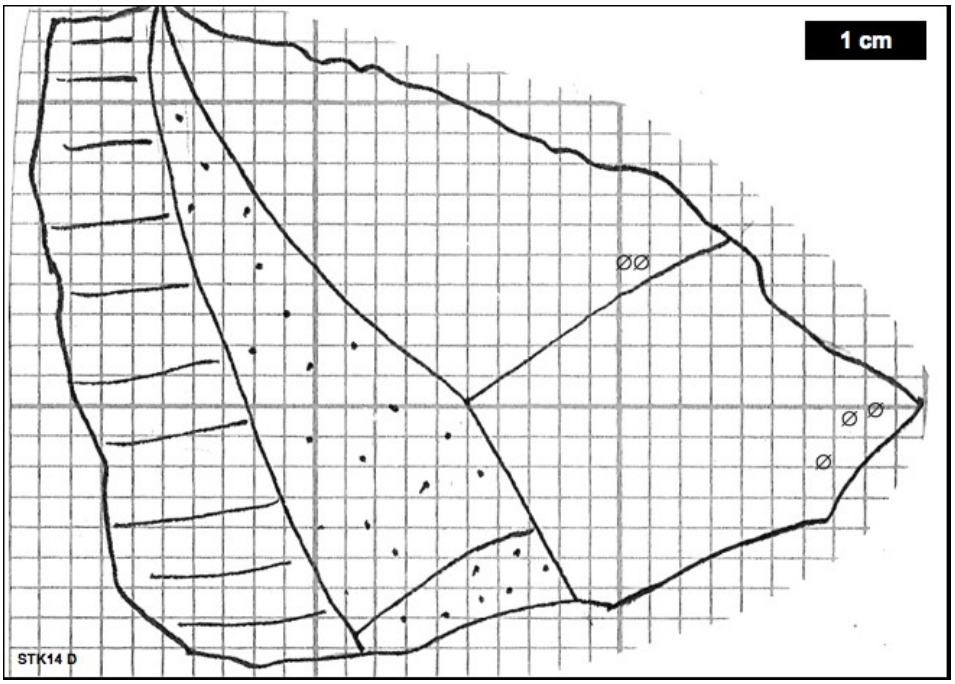


Figure B.66: Distribution map for STK14, dorsal side. After one year no starch residues are preserved outside the cave. Note that use-related plant tissue deposits are still present. For key see Figure B.1.

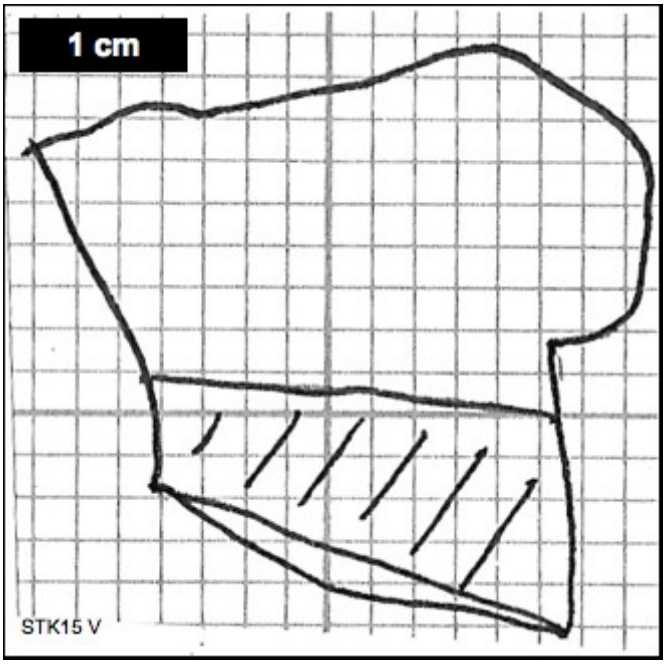


Figure B.67: Distribution map for STK15, ventral side. Even after four weeks almost no starch residues are preserved outside the cave. For key see Figure B.1.

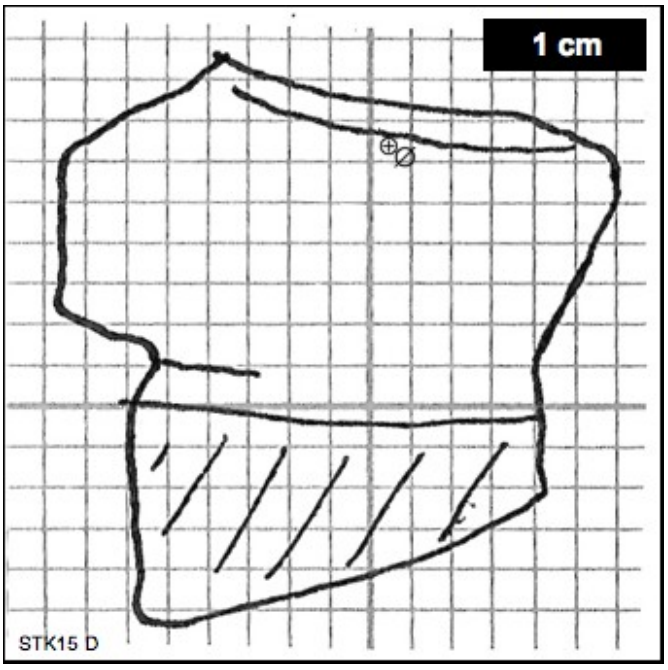


Figure B.68: Distribution map for STK15, dorsal side. Even after four weeks almost no starch residues are preserved outside the cave. For key see Figure B.1.

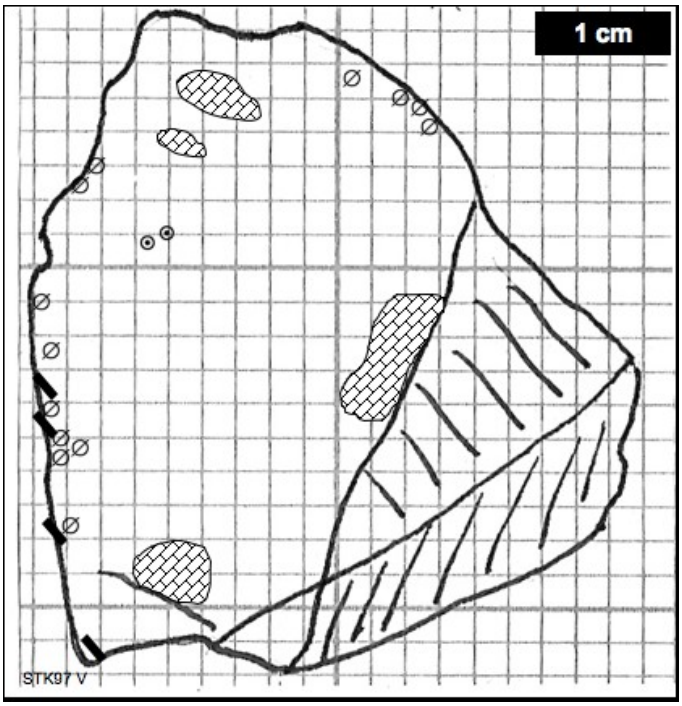


Figure B.69: Distribution map for STK97, ventral side. The use-related plant tissue deposits are confined to the edges. After one year many residues are preserved inside the cave. Note that processing of woody plant materials leaves less residues on a tool's surface than, for example, the processing of a starchy tuber. For key see Figure B.1.

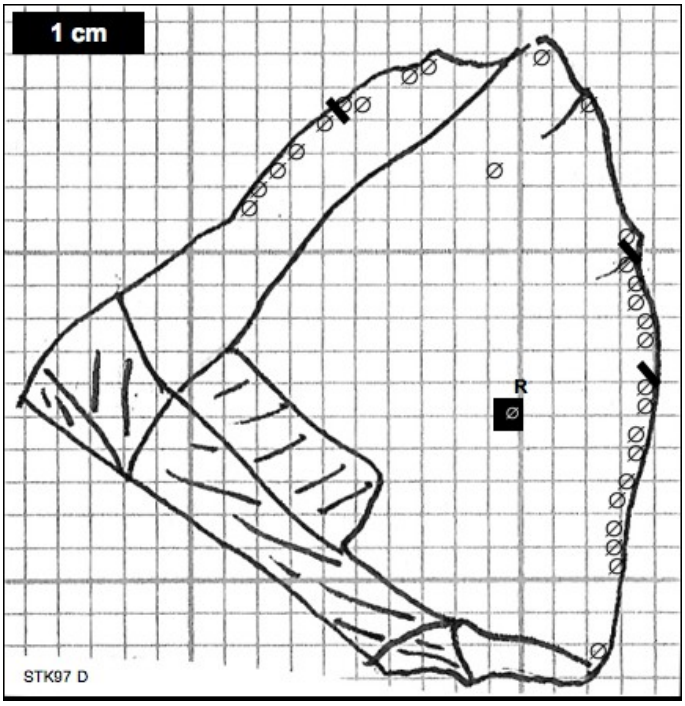


Figure B.70: Distribution map for STK97, dorsal side. The use-related plant tissue deposits are confined to the edges. After one year many residues are preserved inside the cave. Note that processing of woody plant materials leaves less residues on a tool's surface than, for example, the processing of a starchy tuber. For key see Figure B.1.

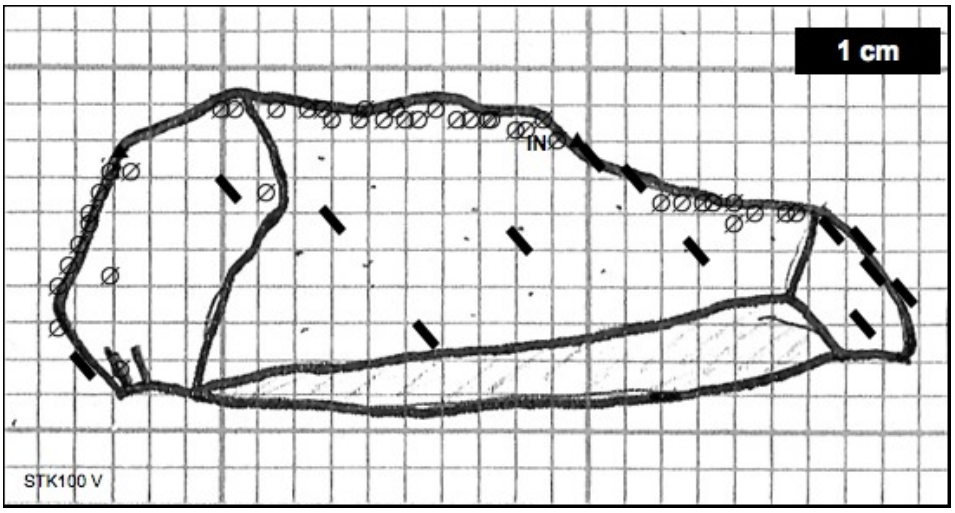


Figure B.71: Distribution map for STK100, ventral side. After four weeks, the preservation of woody plant tissue inside the cave is extremely good. The plant fibres are contaminants. For key see Figure B.1.

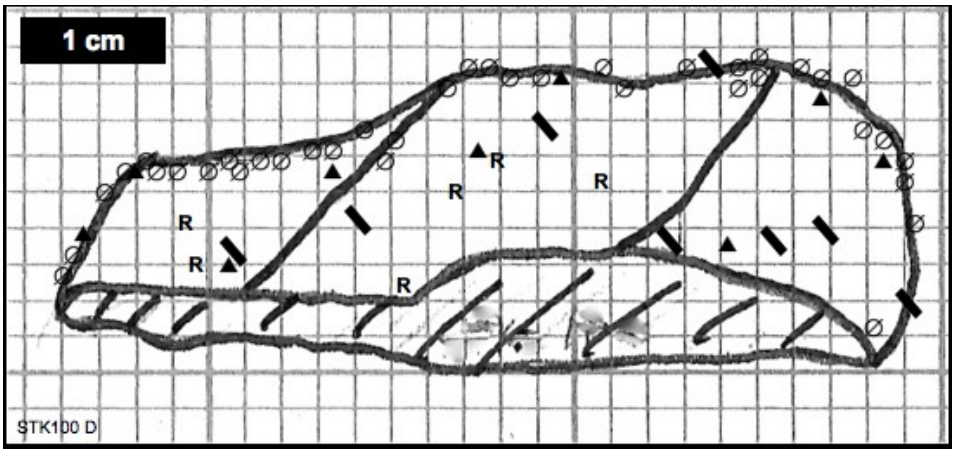


Figure B.72: Distribution map for STK100, dorsal side. After four weeks, the preservation of woody plant tissue inside the cave is extremely good. The plant fibres are contaminants. For key see Figure B.1.

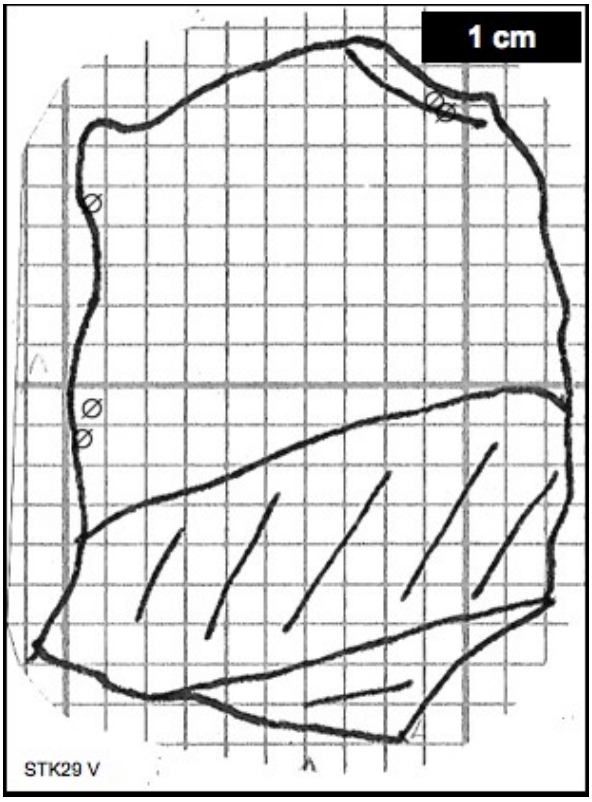


Figure B.73: Distribution map for STK29, ventral side. Outside the cave preservation is considerably less than inside the cave. Only few plant residues are preserved on this tool. For key see Figure B.1.

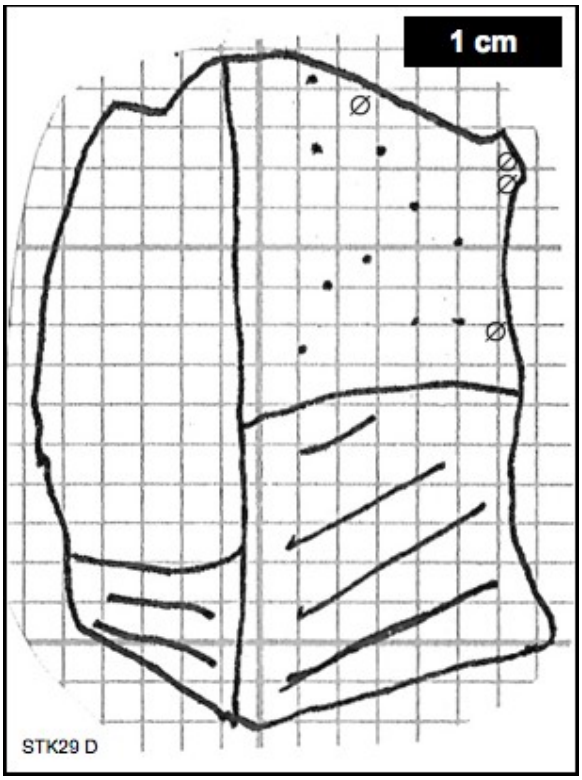


Figure B.74: Distribution map for STK29, dorsal side. Outside the cave preservation is considerably less than inside the cave. Only few plant residues are preserved on this tool. For key see Figure B.1.

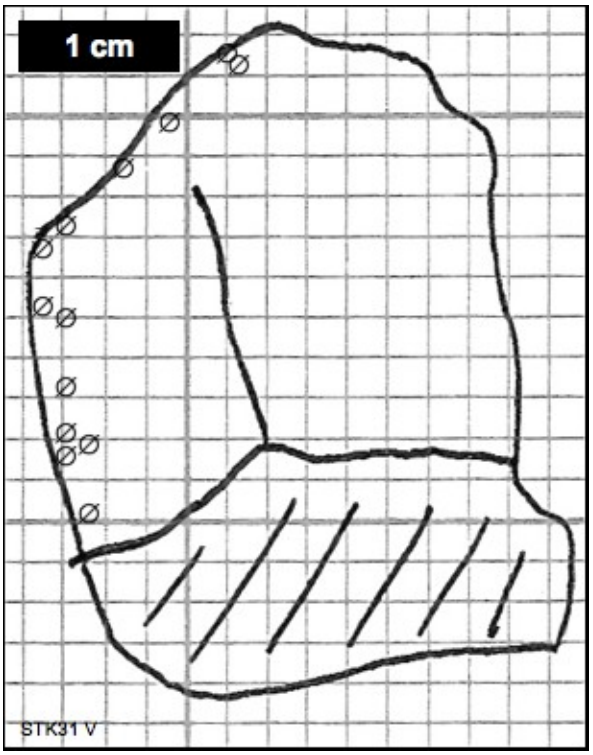


Figure B.75: Distribution map for STK31, ventral side. After four weeks some plant residues preserve outside the cave. For key see Figure B.1.

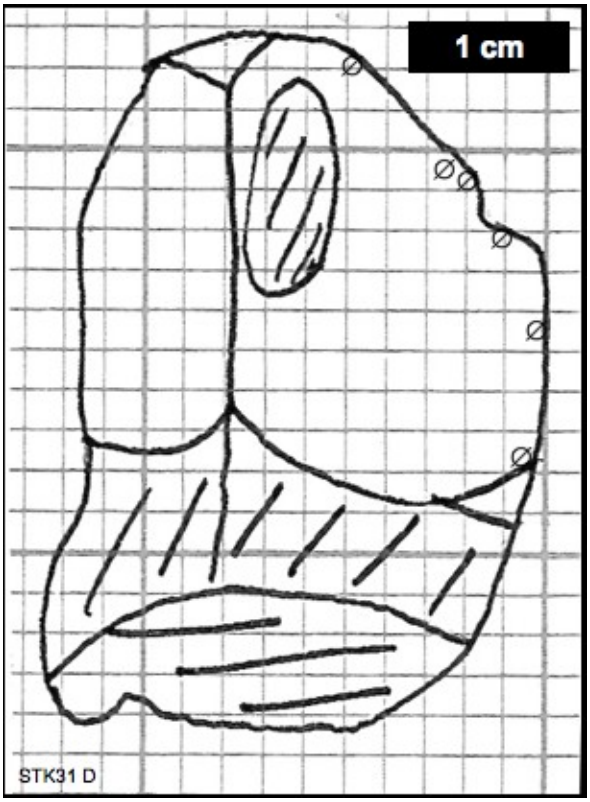


Figure B.76: Distribution map for STK31, dorsal side. After four weeks some plant residues preserve outside the cave. For key see Figure B.1.

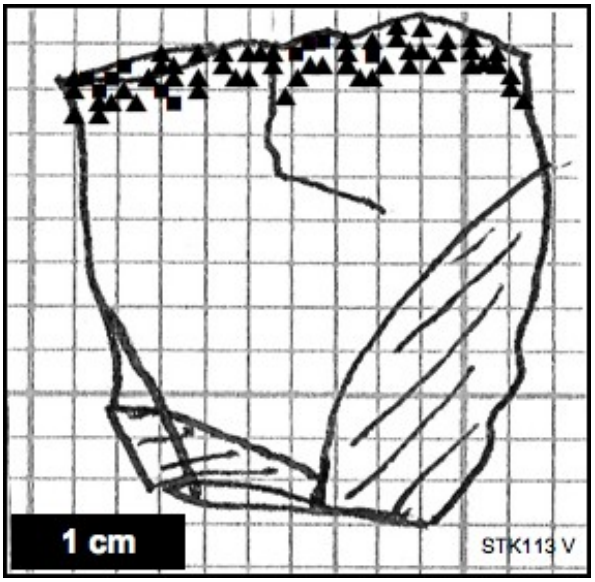


Figure B.77: Distribution map for STK113, ventral side. It is difficult to see the muscle tissue residues through the hyphae deposits on this tool. For key see Figure B.1.

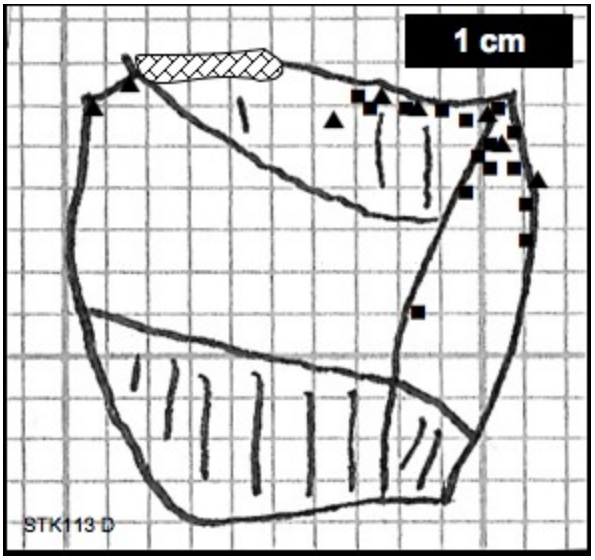


Figure B.78: Distribution map for STK113, dorsal side. Note that the muscle tissue residues are concentrated along the edges. For key see Figure B.1.

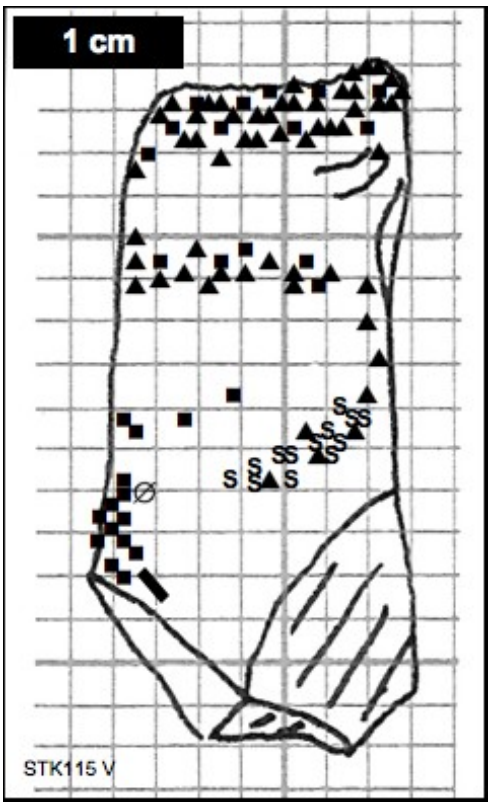


Figure B.79: Distribution map for STK115, ventral side. After four weeks, the muscle tissue residues are well preserved inside the cave. Note that hyphae are abundant on this tool. For key see Figure B.1.

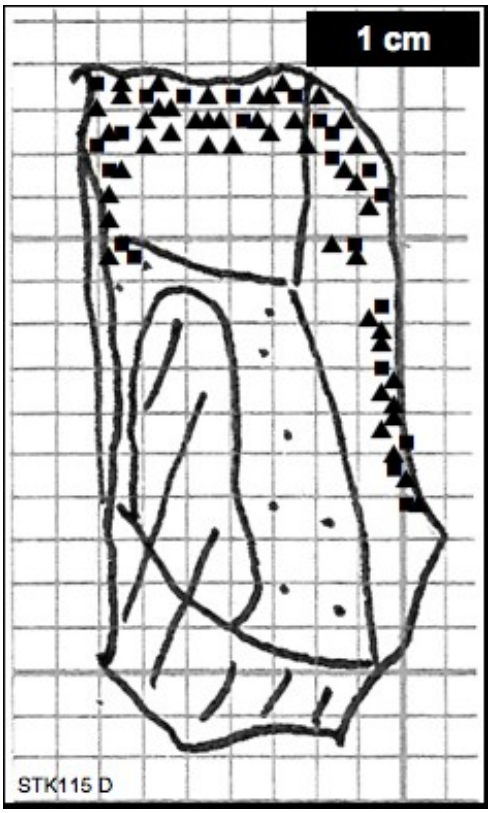


Figure B.80: Distribution map for STK115, dorsal side. After four weeks, the muscle tissue residues are well preserved inside the cave. Note that hyphae are abundant on this tool. For key see Figure B.1.

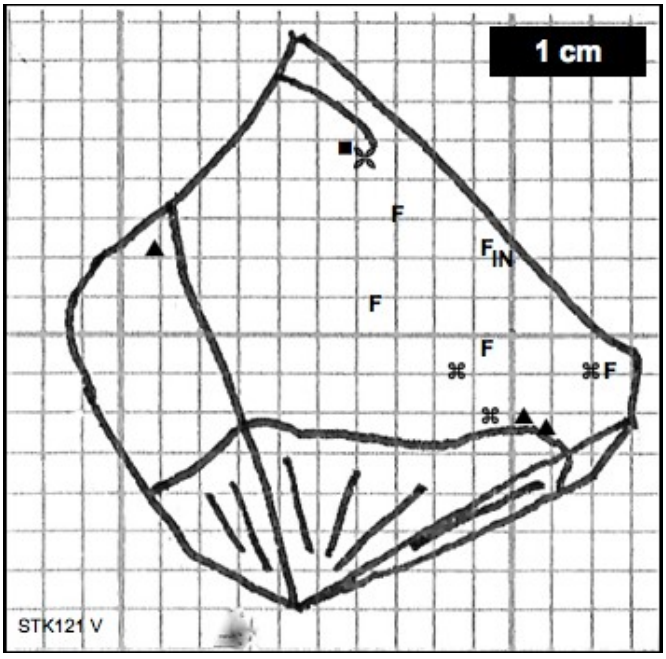


Figure B.81: Distribution map for STK121, ventral side. After one year only a few muscle tissue residues preserve outside the cave. Note that fat on this tool is associated with the muscle tissue. For key see Figure B.1.

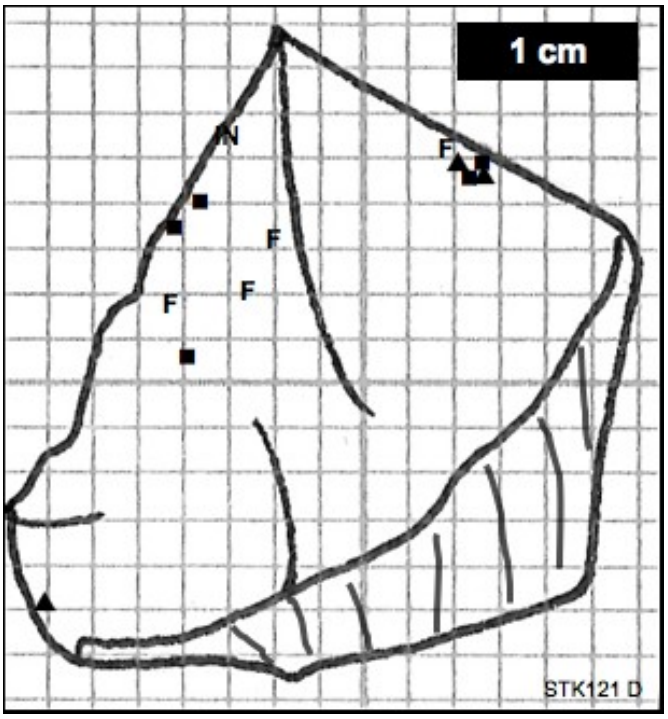


Figure B.82: Distribution map for STK121, dorsal side. After one year only a few muscle tissue residues preserve outside the cave. Note that fat on this tool is associated with the muscle tissue. For key see Figure B.1.

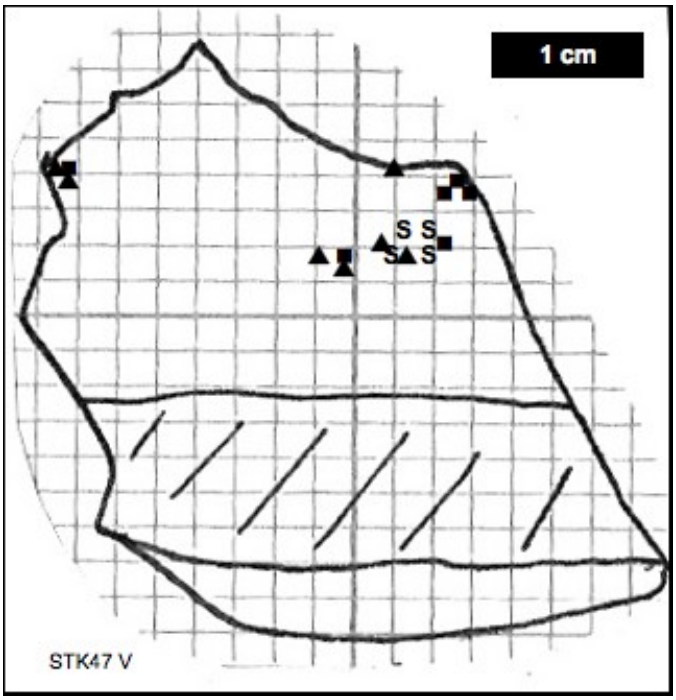


Figure B.83: Distribution map for STK47, ventral side. After four weeks only a few muscle tissue residues preserve outside the cave. For key see Figure B.1.

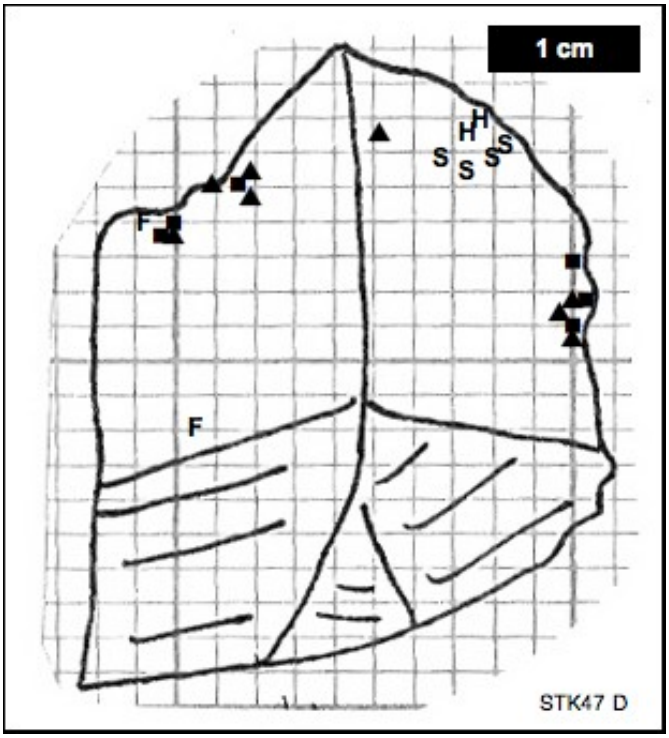


Figure B.84: Distribution map for STK47, dorsal side. After four weeks only a few muscle tissue residues preserve outside the cave. Note the two hairs, which are associated with the muscle tissue residues. For key see Figure B.1.

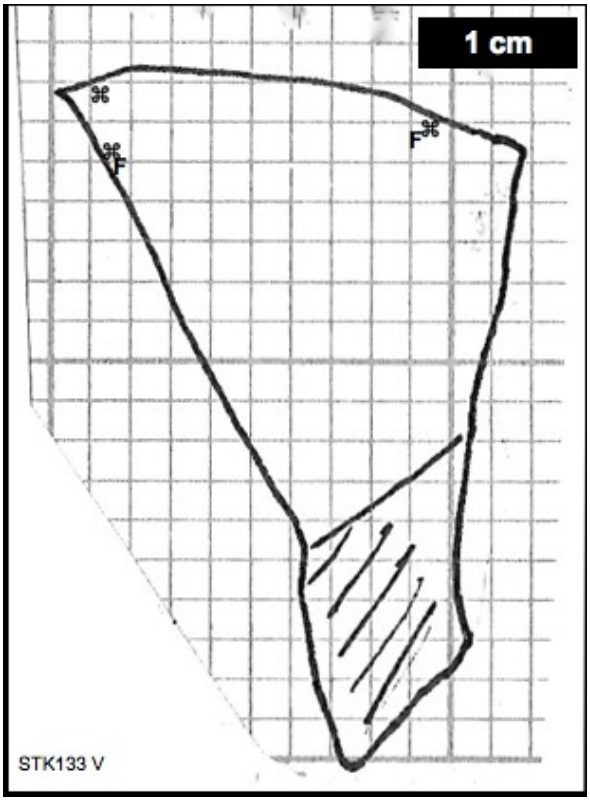


Figure B.85: Distribution map for STK133, ventral side. After one year a few bone residues preserve inside the cave. Note that bone processing leaves considerably less residues on a tool's surface than, for example, processing muscle tissue. For key see Figure B.1.

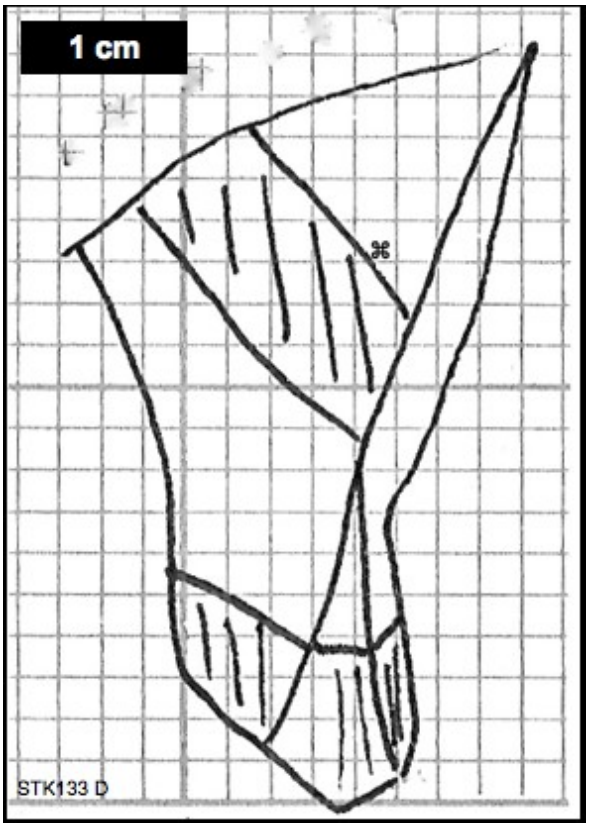


Figure B.86: Distribution map for STK133, dorsal side. After one year a few bone residues preserve inside the cave. Note that bone processing leaves considerably less residues on a tool's surface than, for example, processing muscle tissue. For key see Figure B.1.

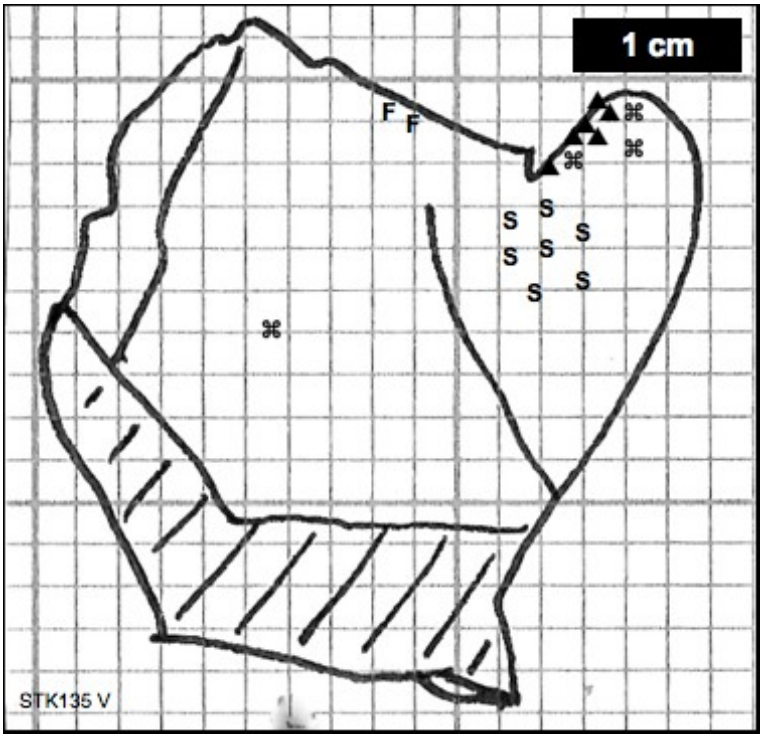


Figure B.87: Distribution map for STK135, ventral side. After four weeks many bone residues are preserved inside the cave. For key see Figure B.1.

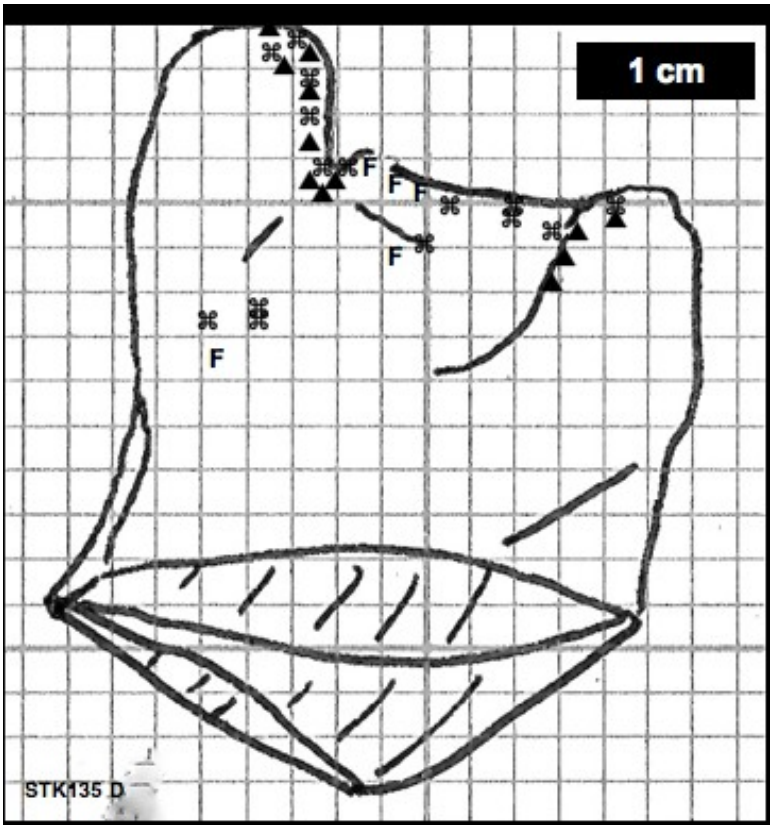


Figure B.88: Distribution map for STK135, dorsal side. After four weeks many bone residues are preserved inside the cave. Note that the bone fat deposit toward the middle of the tool, away from the edge, is probably the result of handling. For key see Figure B.1.

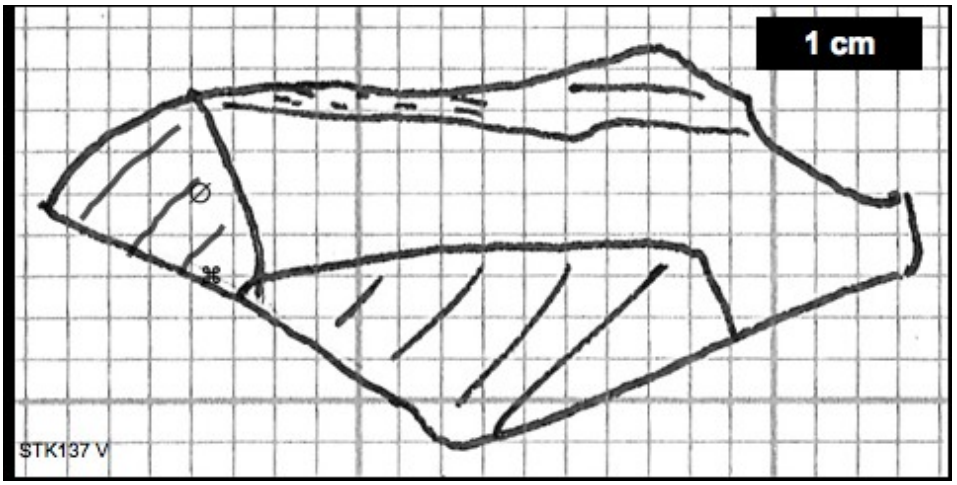


Figure B.89: Distribution map for STK137, ventral side. After one year few bone residues are preserved outside the cave. For key see Figure B.1.

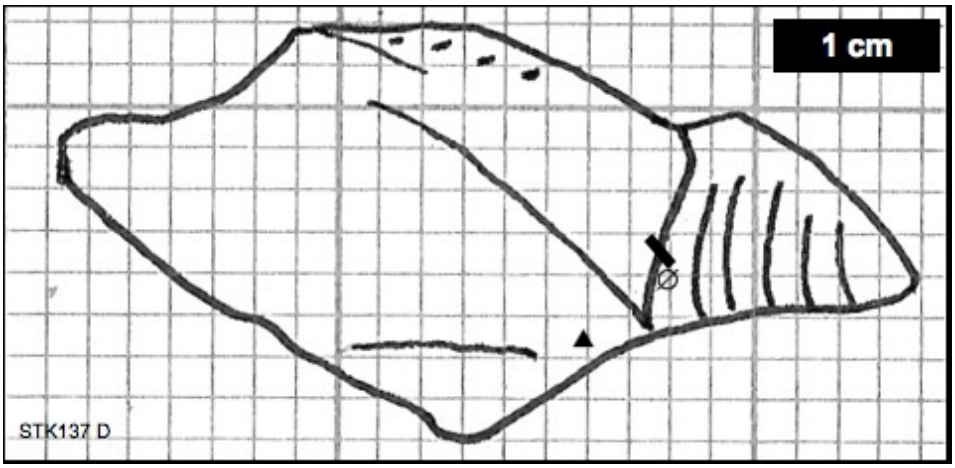


Figure B.90: Distribution map for STK137, dorsal side. After one year few bone residues are preserved outside the cave. For key see Figure B.1.

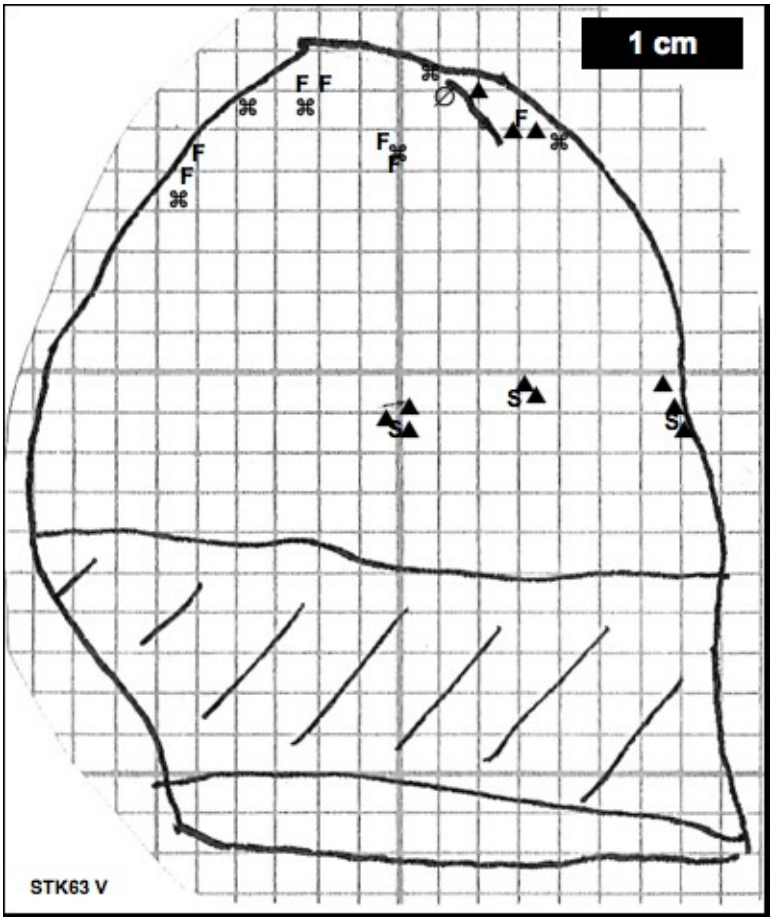


Figure B.91: Distribution map for STK63, ventral side. After four weeks bone is reasonably preserved outside the cave. Note that the fat residues are associated with the bone deposits. For key see Figure B.1.

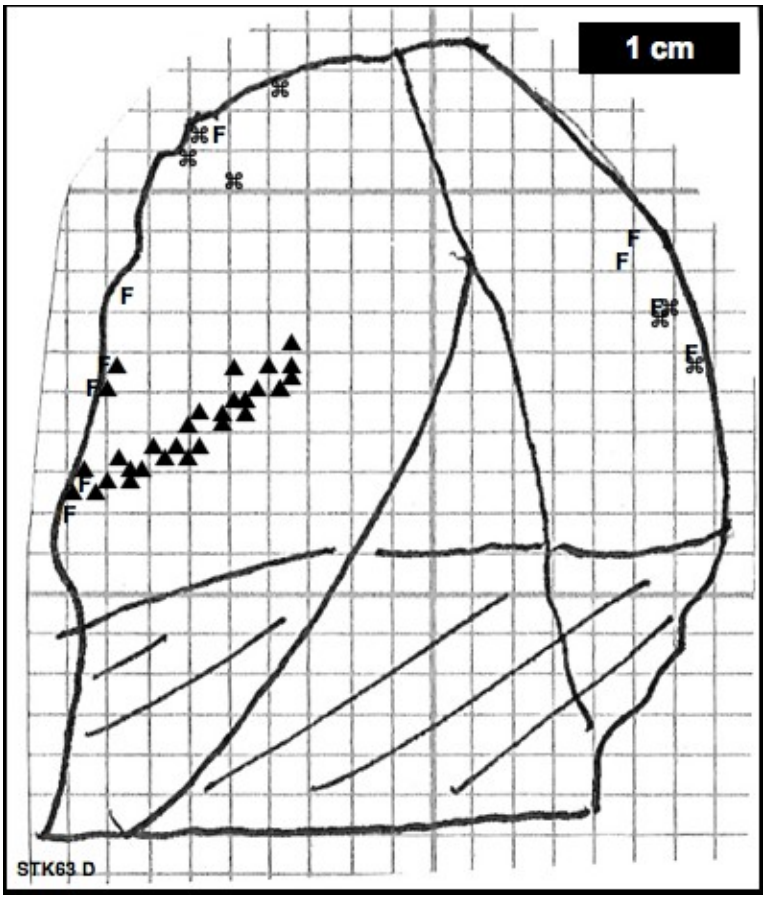


Figure B.92: Distribution map for STK63, dorsal side. After four week bone is reasonably preserved outside the cave. Note that the fat residues are associated with the bone deposits. For key see Figure B.1.

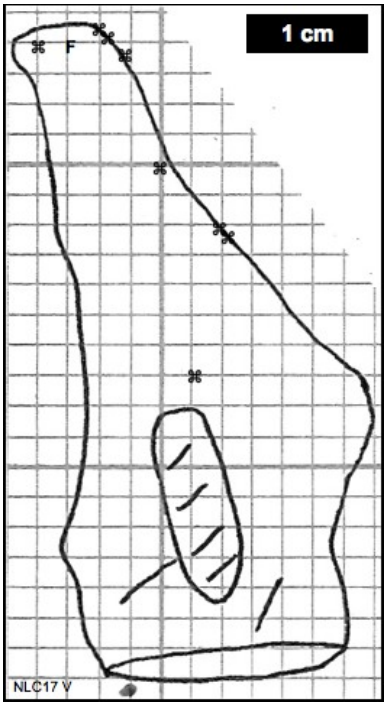


Figure B.93: Distribution map for NLC17, ventral side. Some residues are preserved on the ventral side is reasonable, but there are no fat or bone residues left on the dorsal side. For key see Figure B.1.

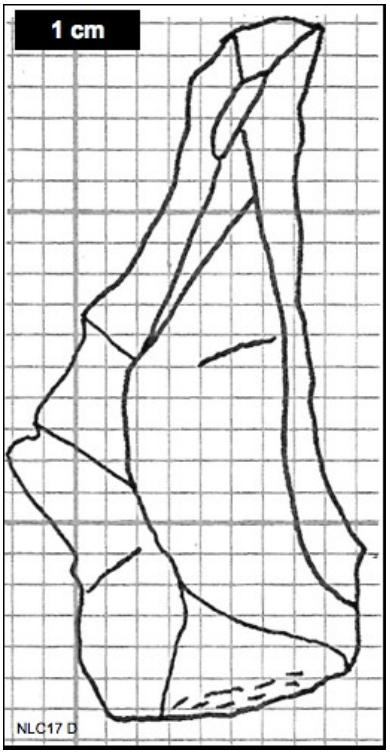


Figure B.94: Distribution map for NLC17, dorsal side. No fat or bone residues left on the dorsal side of this tool. For key see Figure B.1.

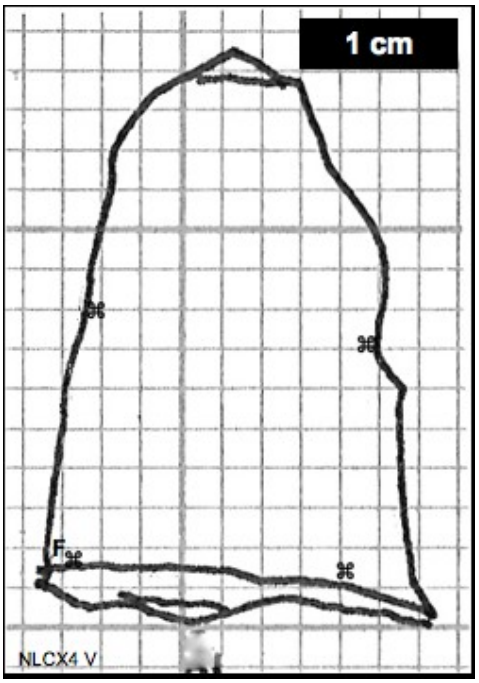


Figure B.95: Distribution map for NLCX4, ventral side. Small bone and fat deposits are preserved on this buried tool. For key see Figure B.1.

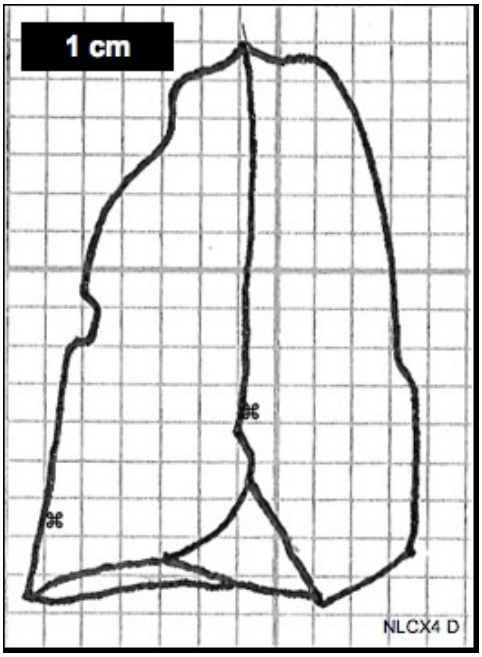


Figure B.96: Distribution map for NLCX4, dorsal side. Small bone and fat deposits are preserved on this buried tool. For key see Figure B.1.

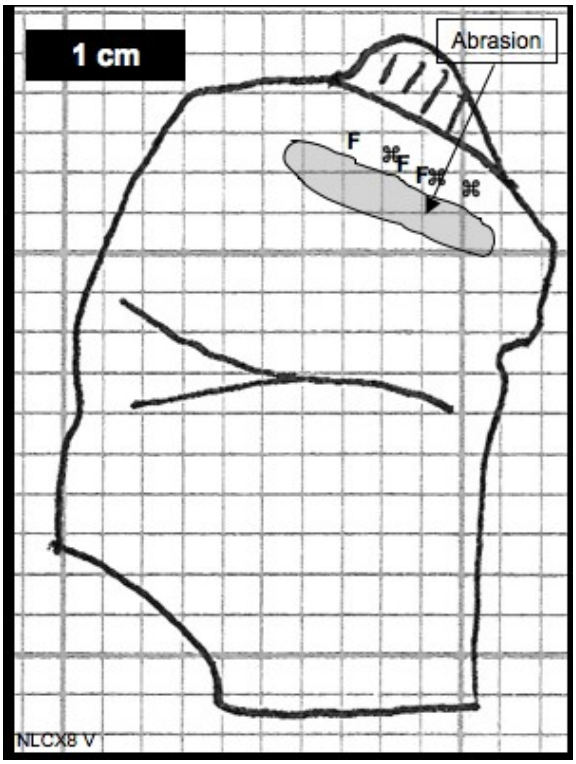


Figure B.97: Distribution map for NLCX8, ventral side. A thick fat and bone deposit is preserved on the ventral side of this exposed tool. For key see Figure B.1.

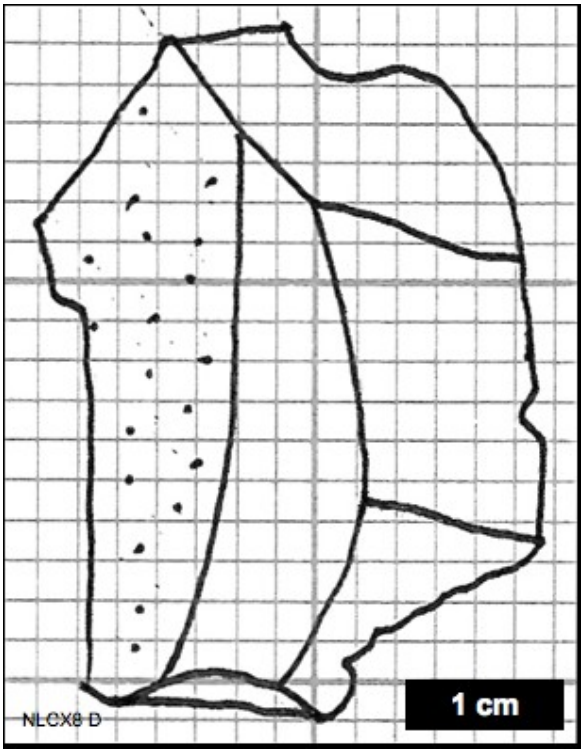


Figure B.98: Distribution map for NLCX8, dorsal side. A thick fat and bone deposit is preserved on the ventral side of this exposed tool; see Figure B.97. For key see Figure B.1.

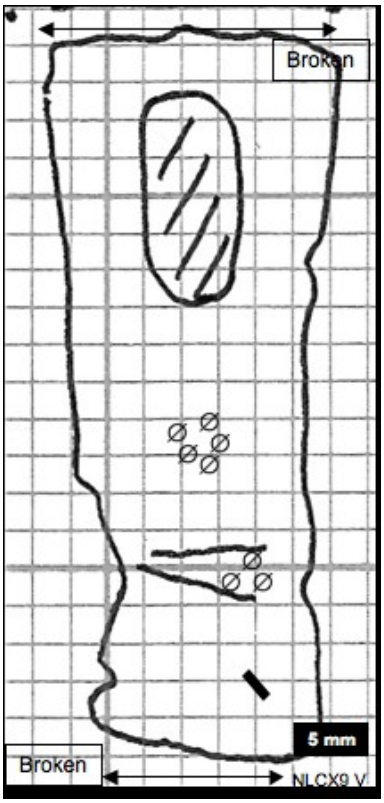


Figure B.99: Distribution map for NLCX9, ventral side. No starch grains are preserved on this exposed tool, but many use-related plant tissue deposits are preserved. This demonstrates the need for contextual analysis. For key see Figure B.1.

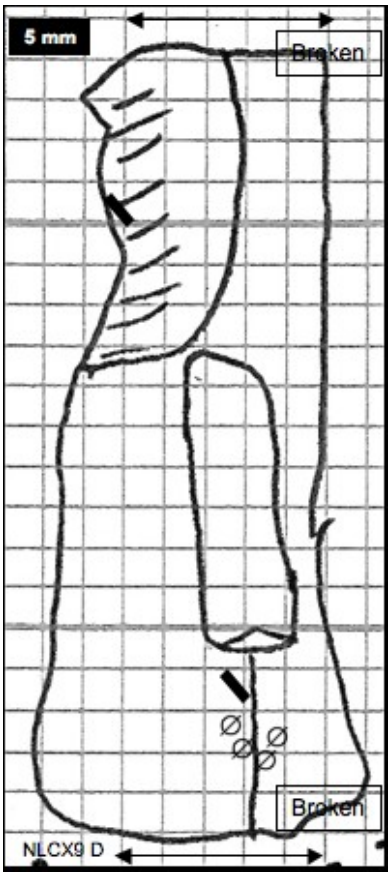


Figure B.100: Distribution map for NLCX9, dorsal side. No starch grains are preserved on this exposed tool, but many use-related plant tissue deposits are preserved. This demonstrates the need for contextual analysis. For key see Figure B.1.

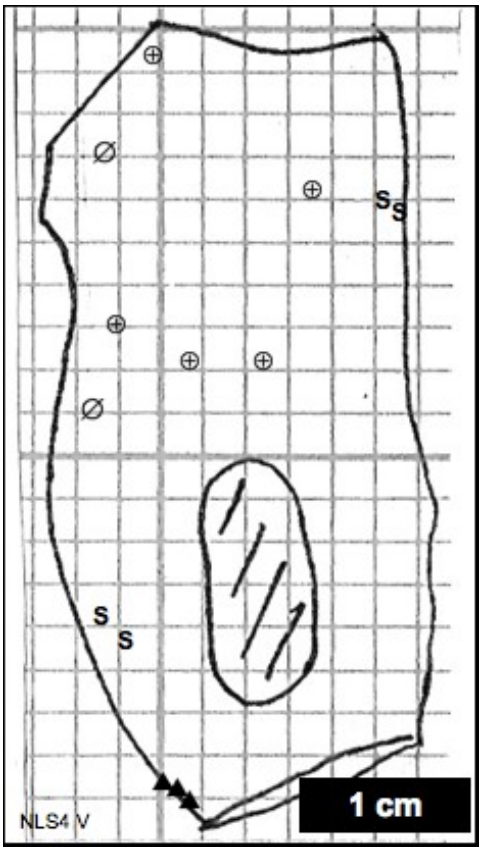


Figure B.101: Distribution map for NLS4, ventral side. Some starch is preserved on this tool. The plant tissue deposits are associated with the starch residues. For key see Figure B.1.

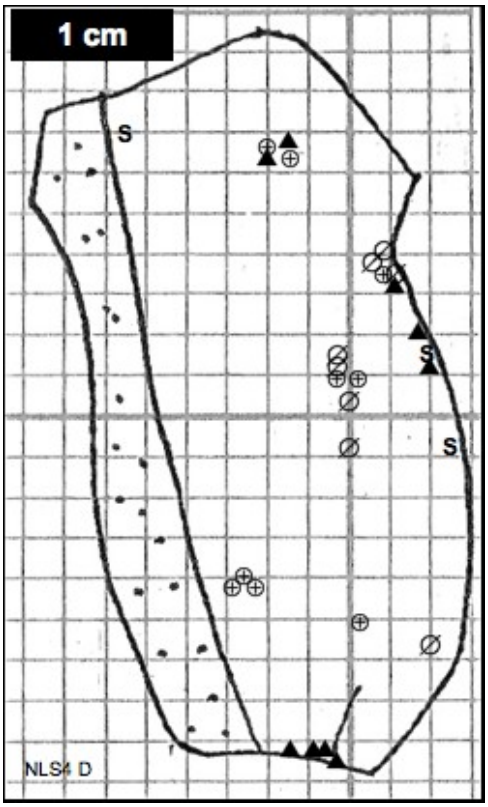


Figure B.102: Distribution map for NLS4, dorsal side. On this exposed tool starch preservation is reasonable. The plant tissue deposits are associated with the starch residues. For key see Figure B.1.

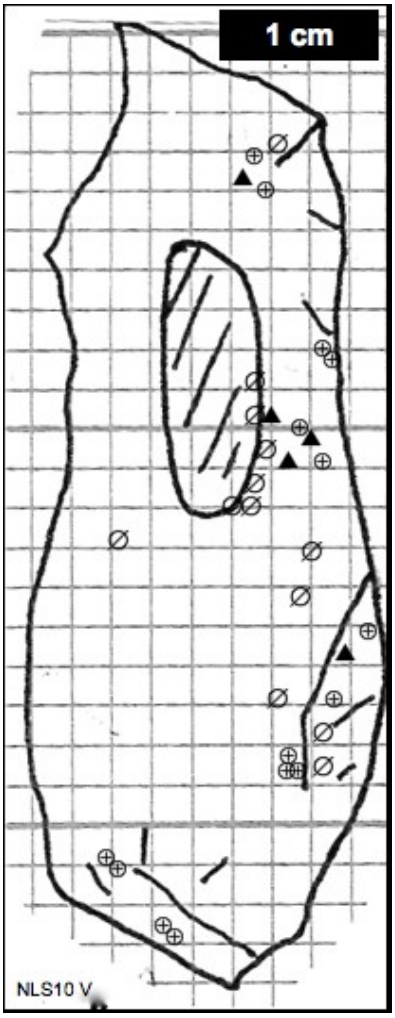


Figure B.103: Distribution map for NLS10, ventral side. Some starch is preserved on this buried tool. The plant tissue deposits are associated with the starch residues. For key see Figure B.1.

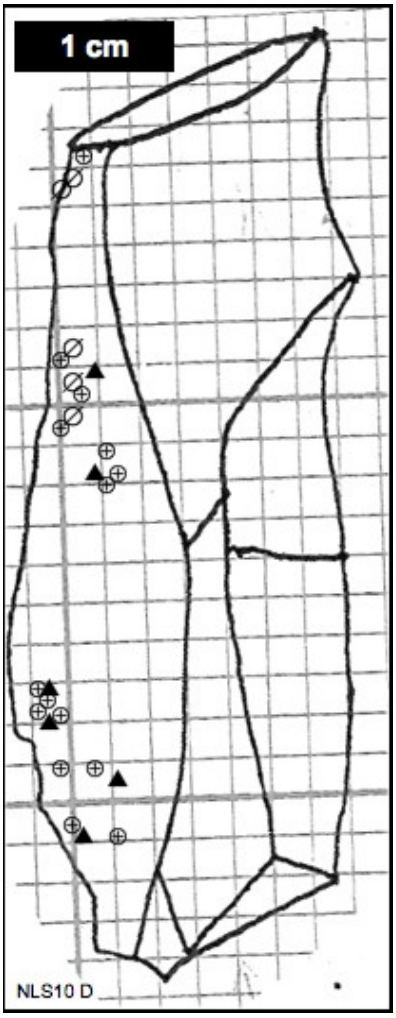


Figure B.104: Distribution map for NLS10, dorsal side. Some starch is preserved on this buried tool. The plant tissue deposits are associated with the starch residues. For key see Figure B.1.

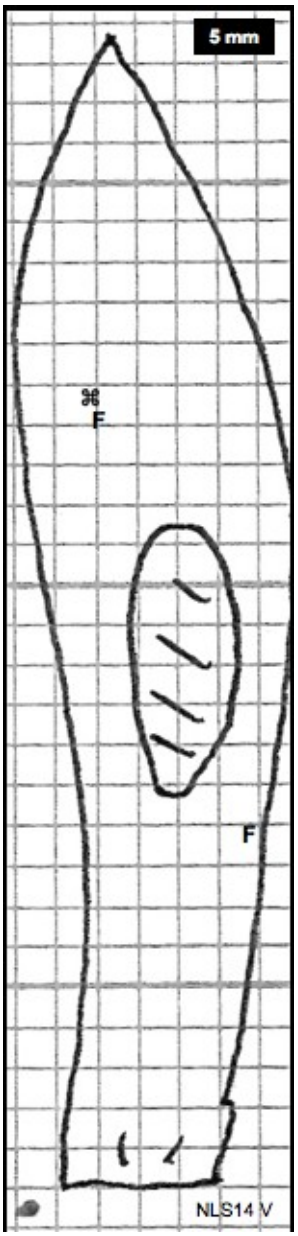


Figure B.105: Distribution map for NLS14, ventral side. Only a few bone and fat residues are preserved on this exposed tool. For key see Figure B.1.

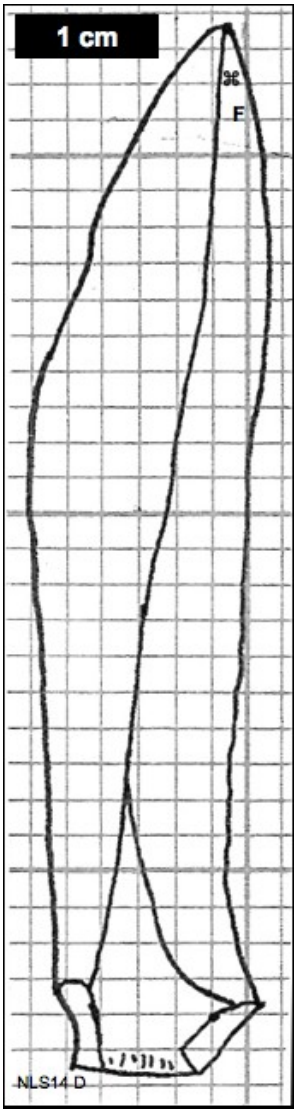


Figure B.106: Distribution map for NLS14, dorsal side. Only a few bone and fat residues are preserved on this exposed tool. For key see Figure B.1.

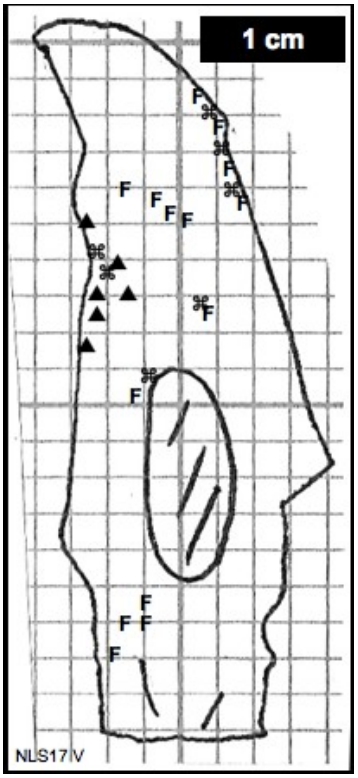


Figure B.107: Distribution map for NLS17, ventral side. The ventral side preserves many bone and fat residues, but the dorsal side just two fat deposits. For key see Figure B.1.

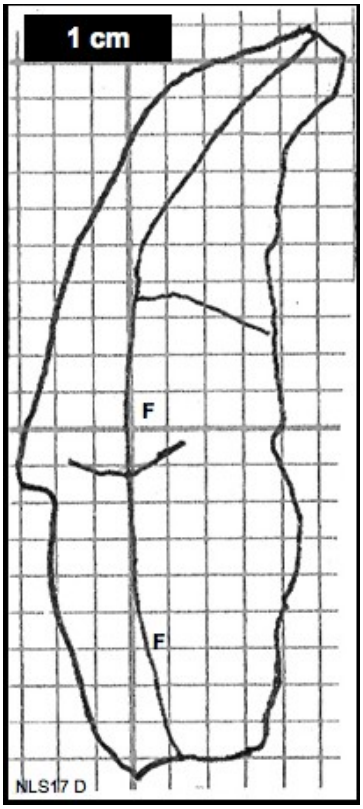


Figure B.108: Distribution map for NLS17, dorsal side. The ventral side preserves many bone and fat residues, but the dorsal side just two fat deposits. For key see Figure B.1.

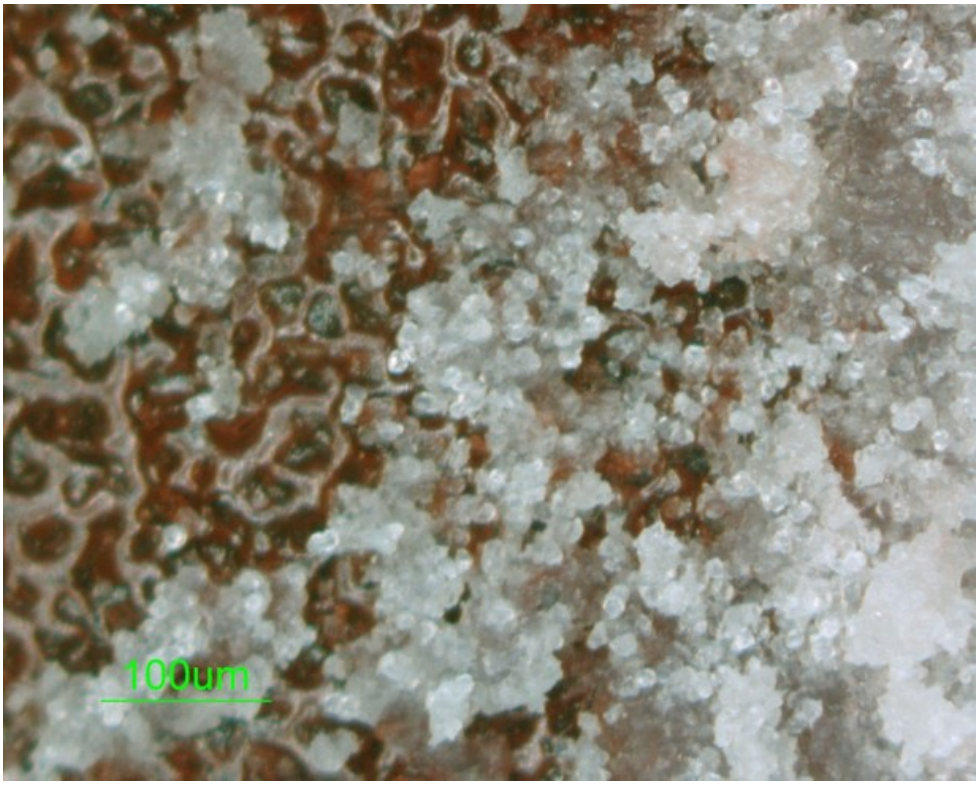


Figure B.109: Fat on NLSF13 before deposition. Note the globular shape of the fat cells. Magnification 10.

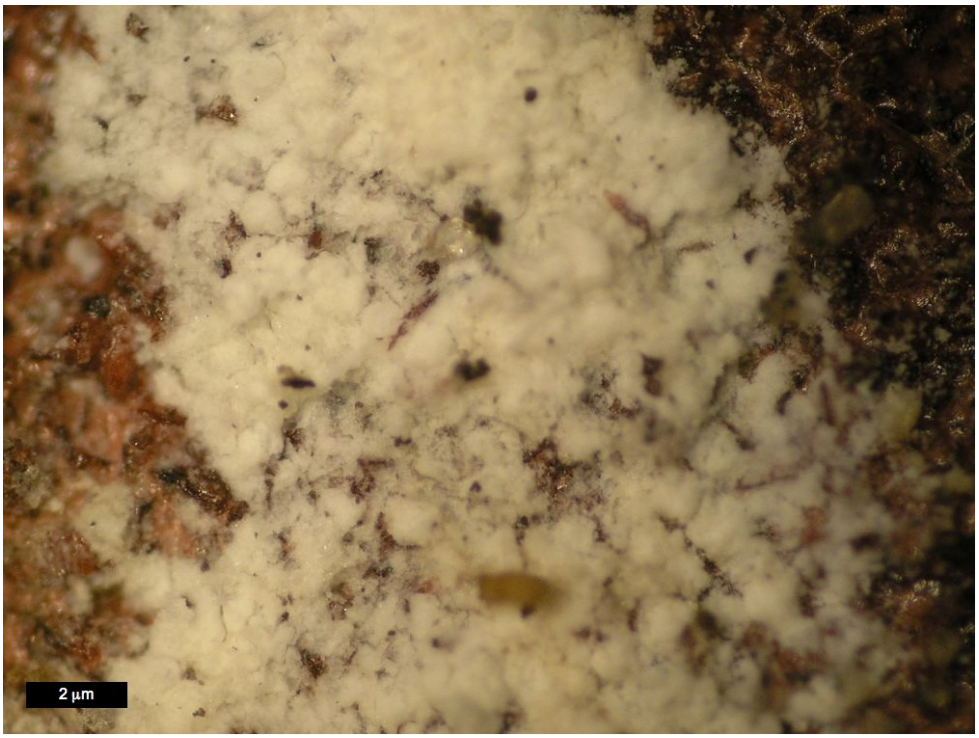


Figure B.110: First hyphae growing on a fat deposit on NLSF10. The deposit has a woolly feel. Magnification 50.

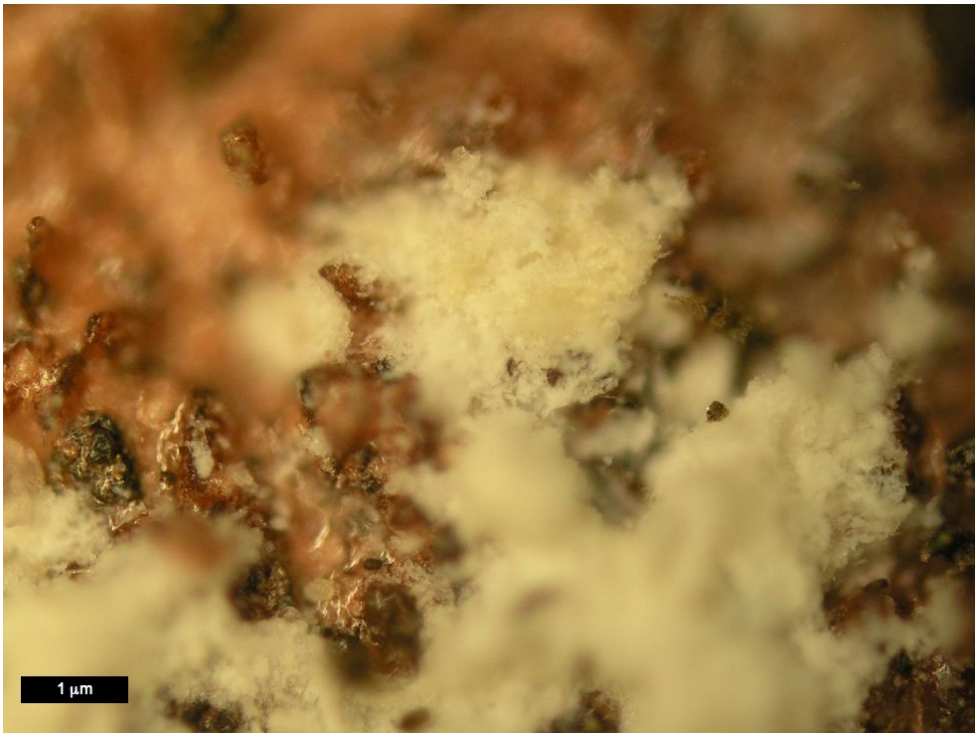


Figure B.111: First hyphae growing on a fat deposit on NLSF9. Magnification 100.

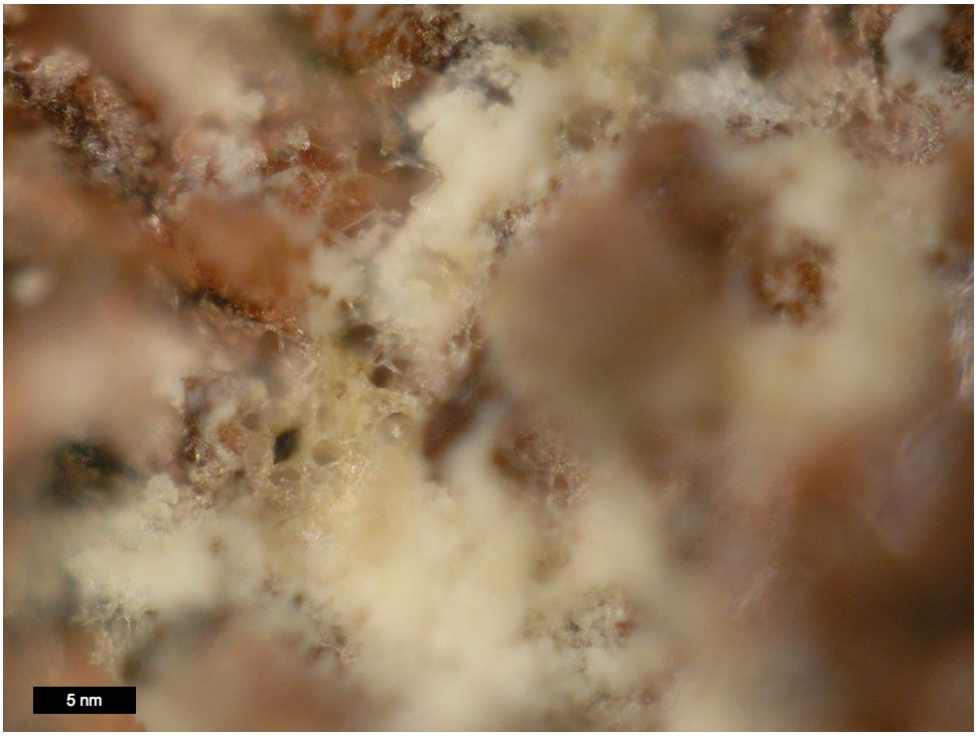


Figure B.112: More hyphae growing on a fat deposit on NLSF12. Now the thick gelatine hyphae strands are visible. Magnification 200.

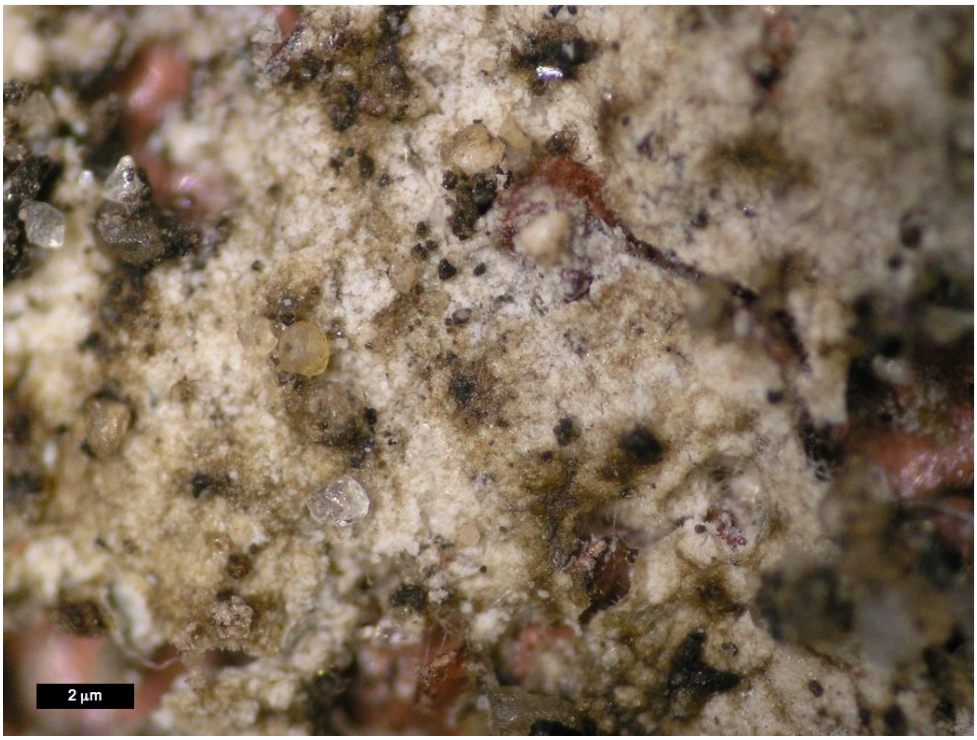


Figure B.113: Hyphae completely dominate a fat deposit on NLSF15. Note the black spore deposits. Magnification 50.

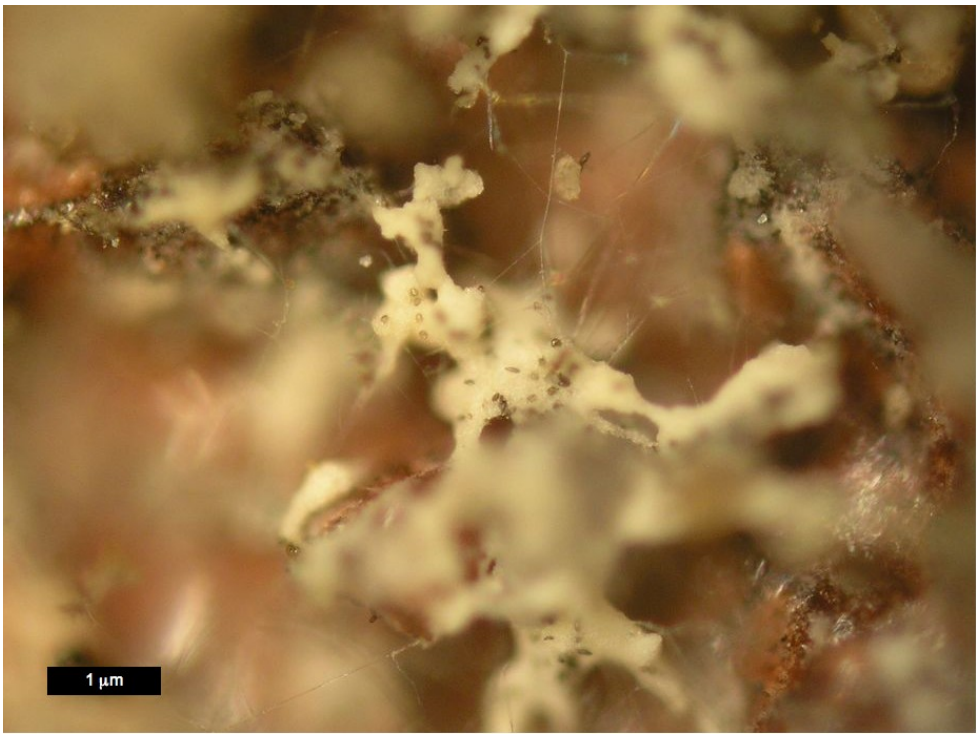


Figure B.114: Hyphae growing on fat on NLSF22. Magnification 100.



Figure B.115: Hyphae on fat on NLSF22. Note that the fat deposit become lumpier and smaller. Magnification 50.

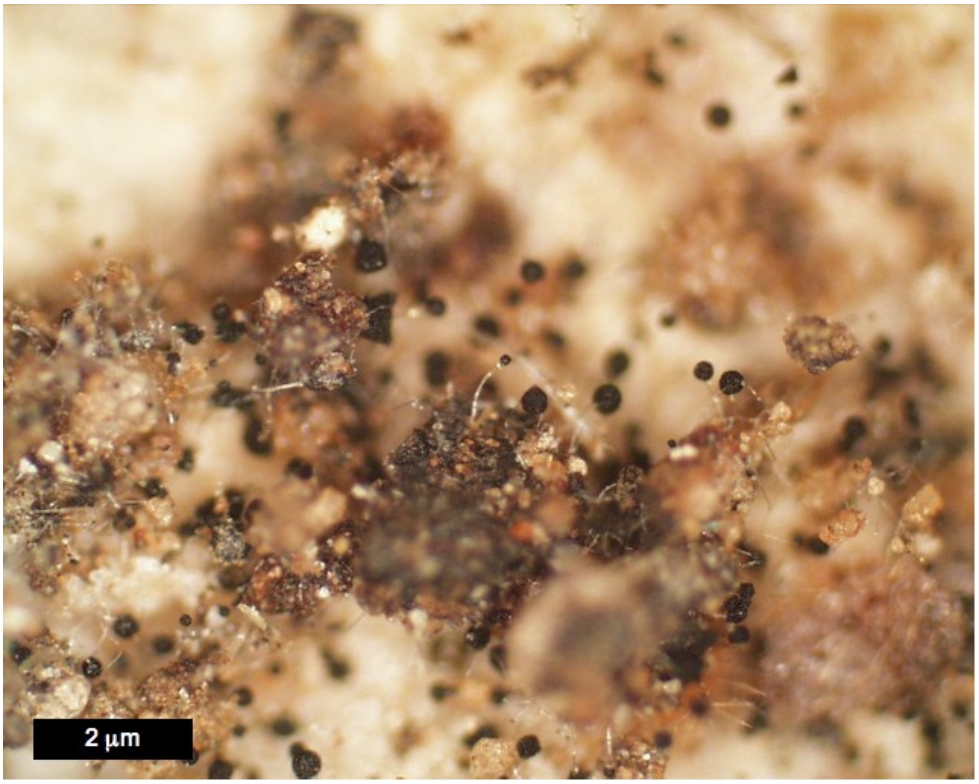


Figure B.116: Spores developing on STK7; these are fungi fruiting bodies. Magnification 50.

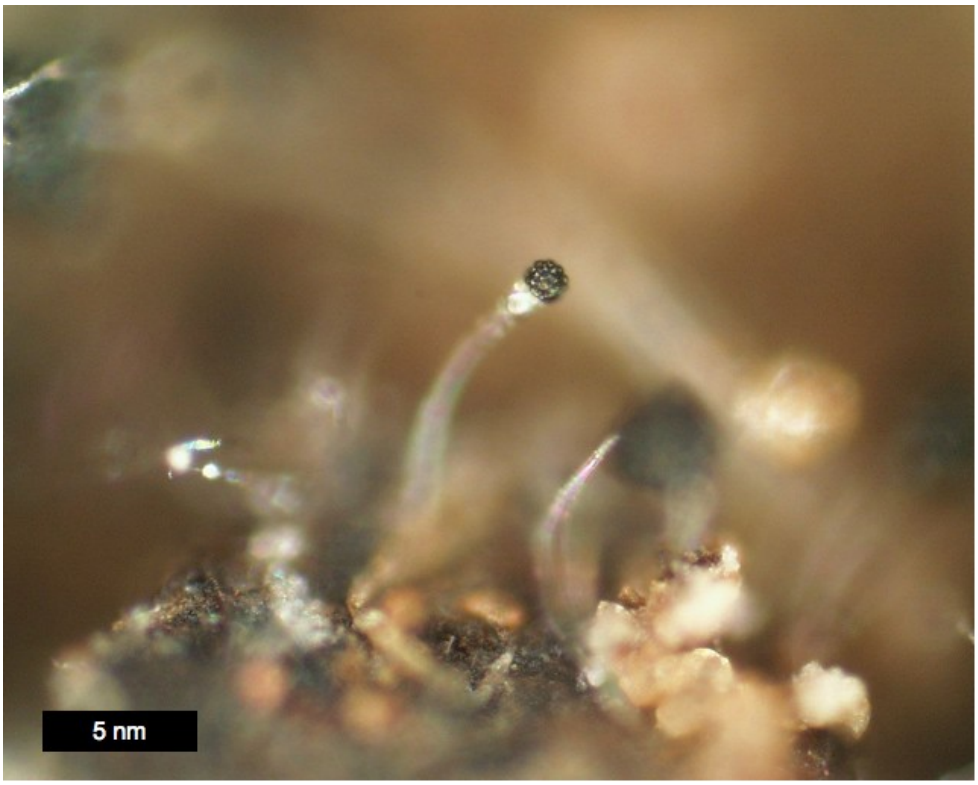


Figure B.117: Spores developing on STK7. This is a fruiting body. Magnification 200.

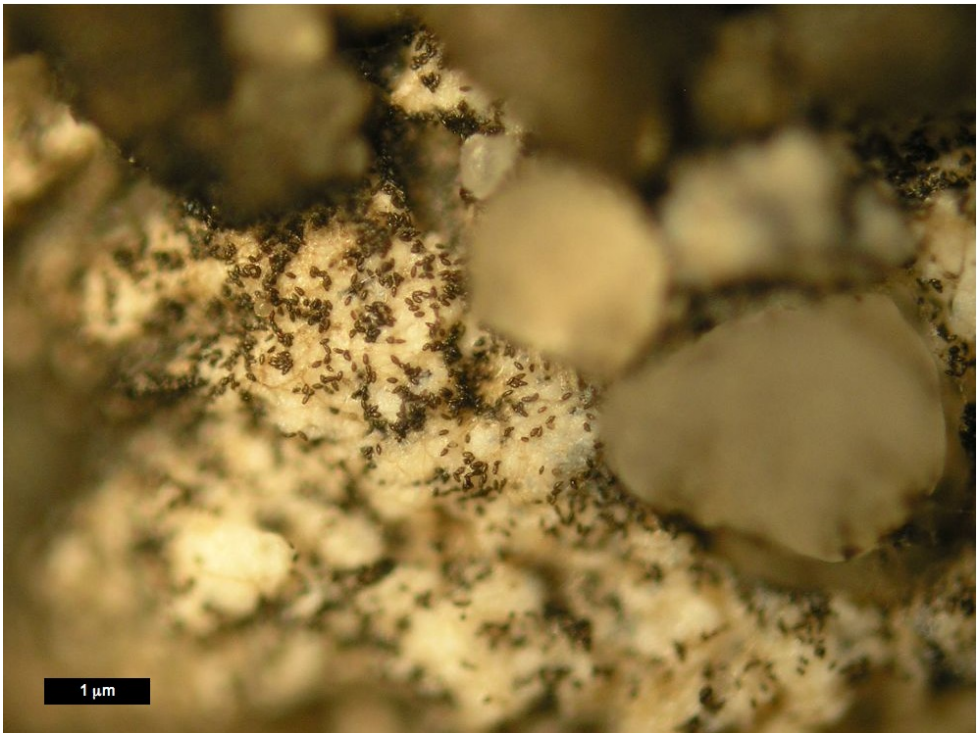


Figure B.118: Hyphae and black spores on fat on NLSF22. Magnification 100.

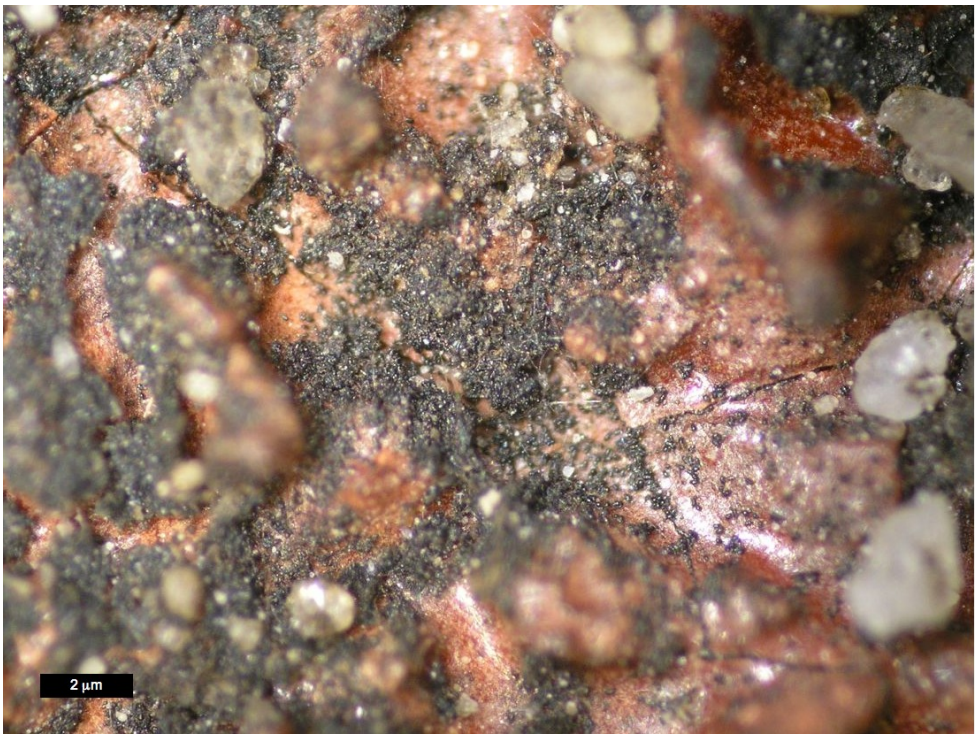


Figure B.119: A thick deposit of spores on NLSF23. Magnification 50.

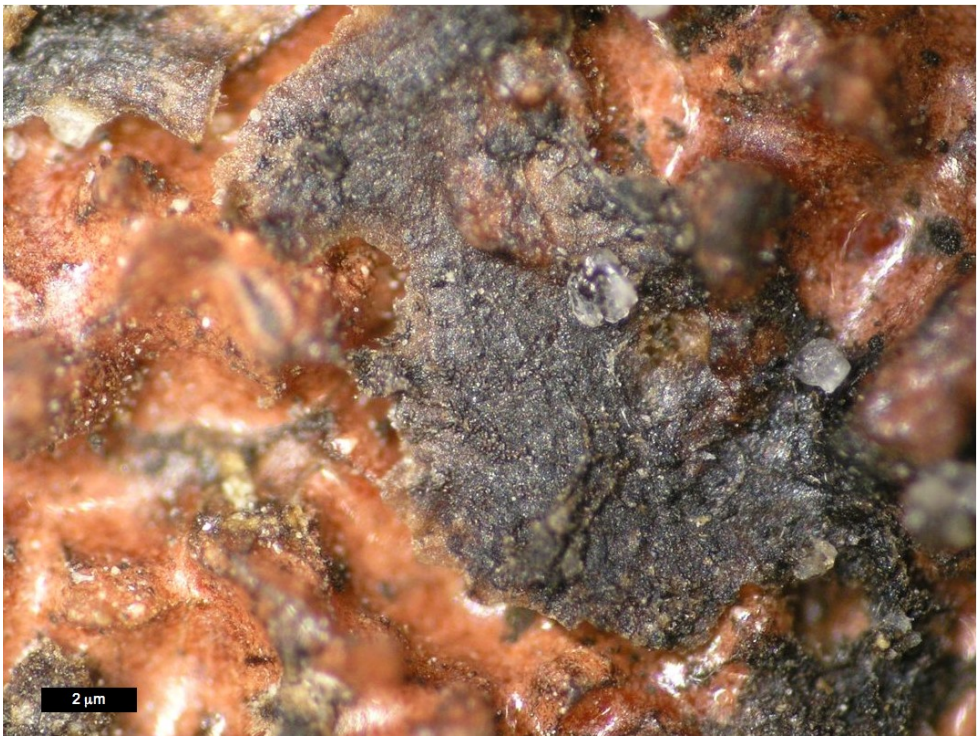


Figure B.120: A thick, dark and consolidated spores/hyphae layer on NLSF19. Magnification 50.

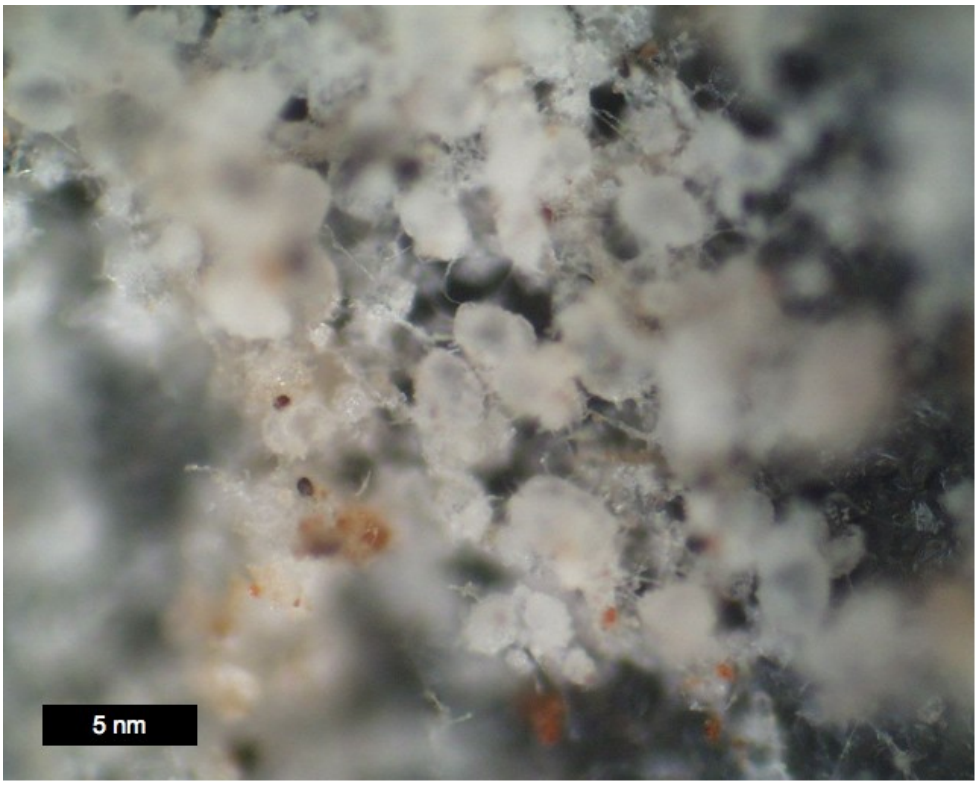


Figure B.121: Hyphae nourishing on starch grains on STK84. Note that the starch grains have become opaque. Magnification 200.

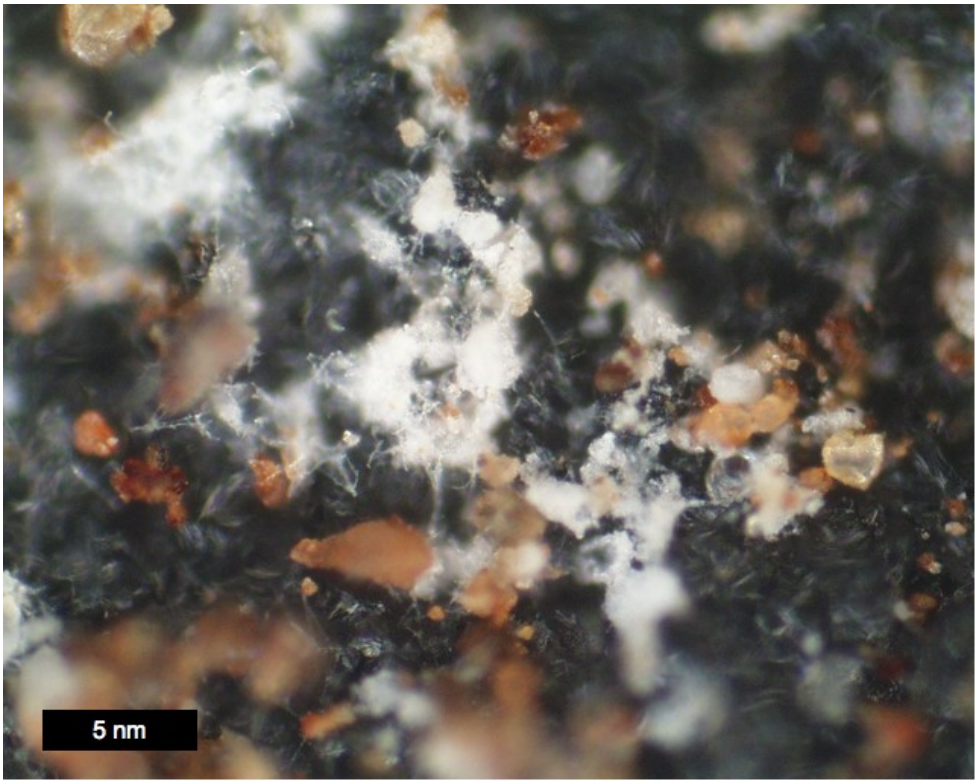


Figure B.122: Hyphae incorporating starch grains on STK88. Magnification 200.



Figure B.123: Hyphae incorporating starch grains on STK88. Once the grains are encapsulated, it is difficult to distinguish them from the hyphae. Magnification 200.

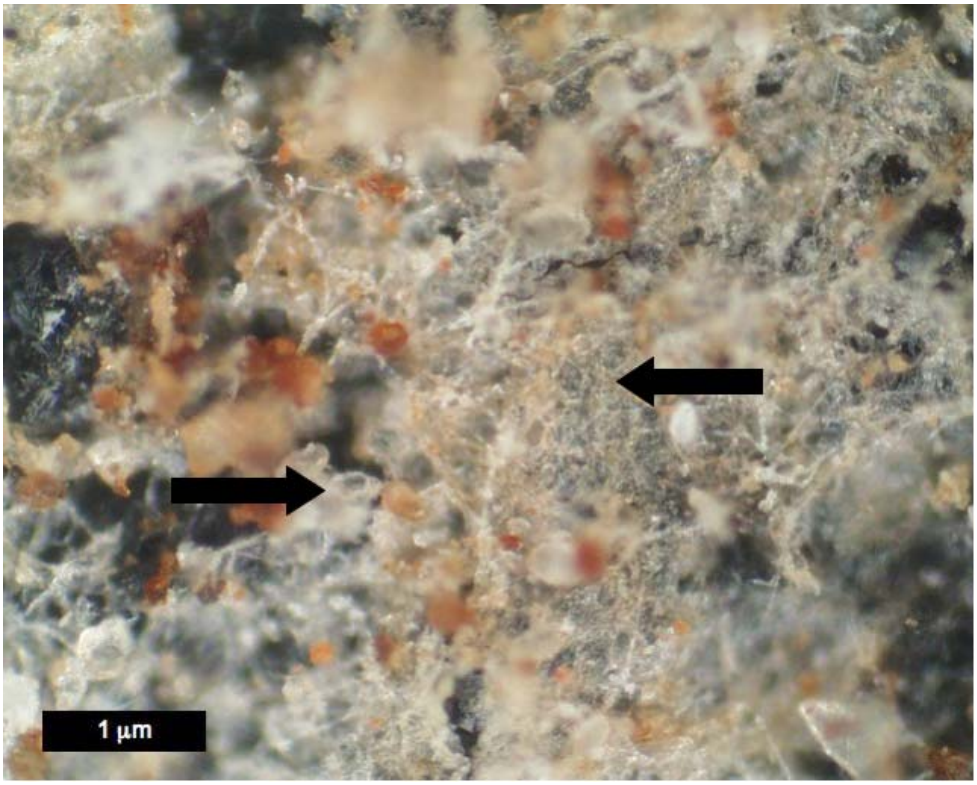


Figure B.124: Hyphae dominating a starch grain deposit on STK88. The starch grains (left arrow) are almost no longer visible and the hyphae deposit (right arrow) resembles animal tissue. Magnification 100.



Figure B.125: Damaged opaque starch grains with hyphae. Magnification 200.

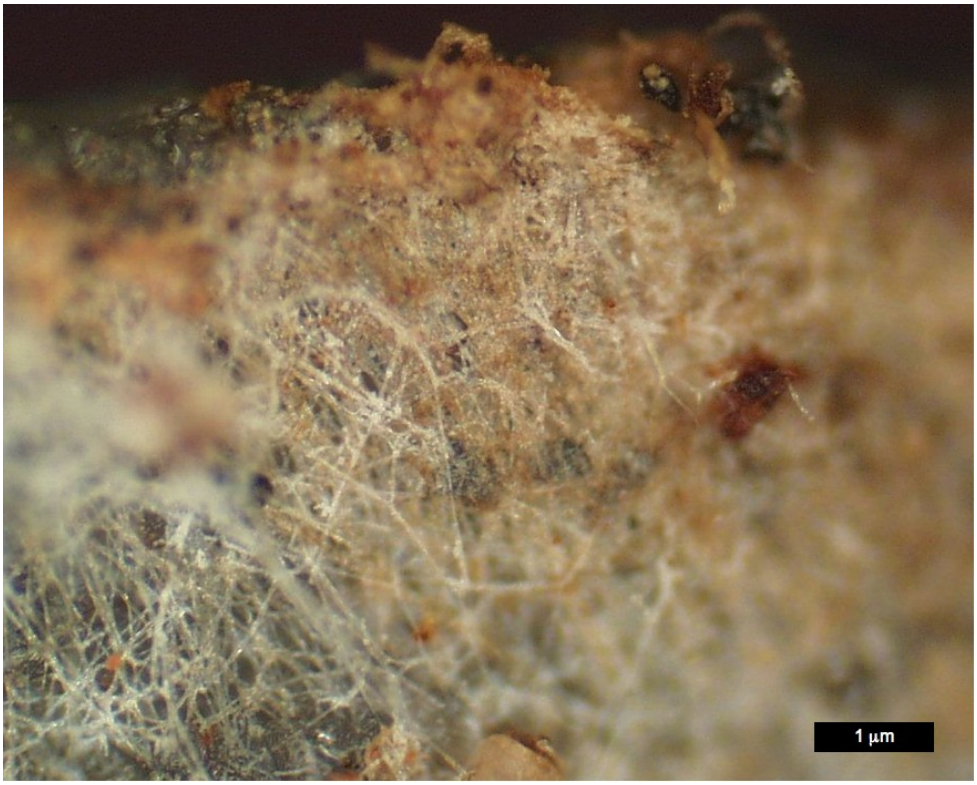


Figure B.126: Hyphae growing on woody plant tissue. Magnification 100.

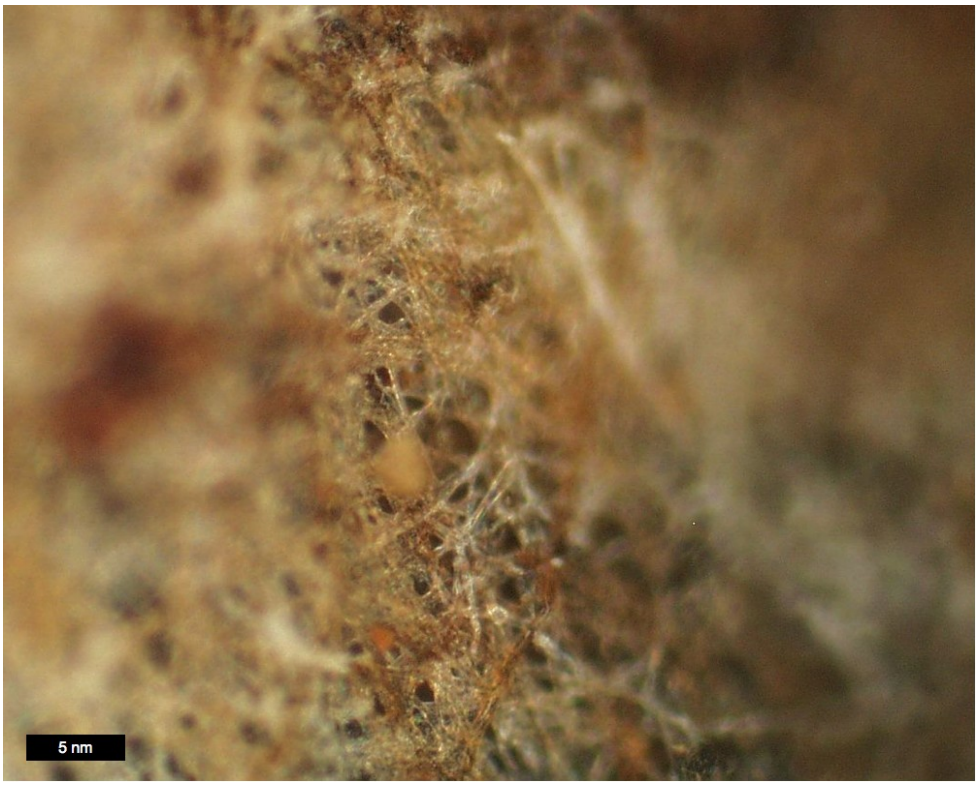


Figure B.127: Hyphae on woody plant tissue. The plant tissue is almost not visible. Magnification 200.



Figure B.128: A plant fibre, which resembles a connective fibre. The fibre is opaque and has a fatty feel, which is typical for connective fibres. Magnification 200.

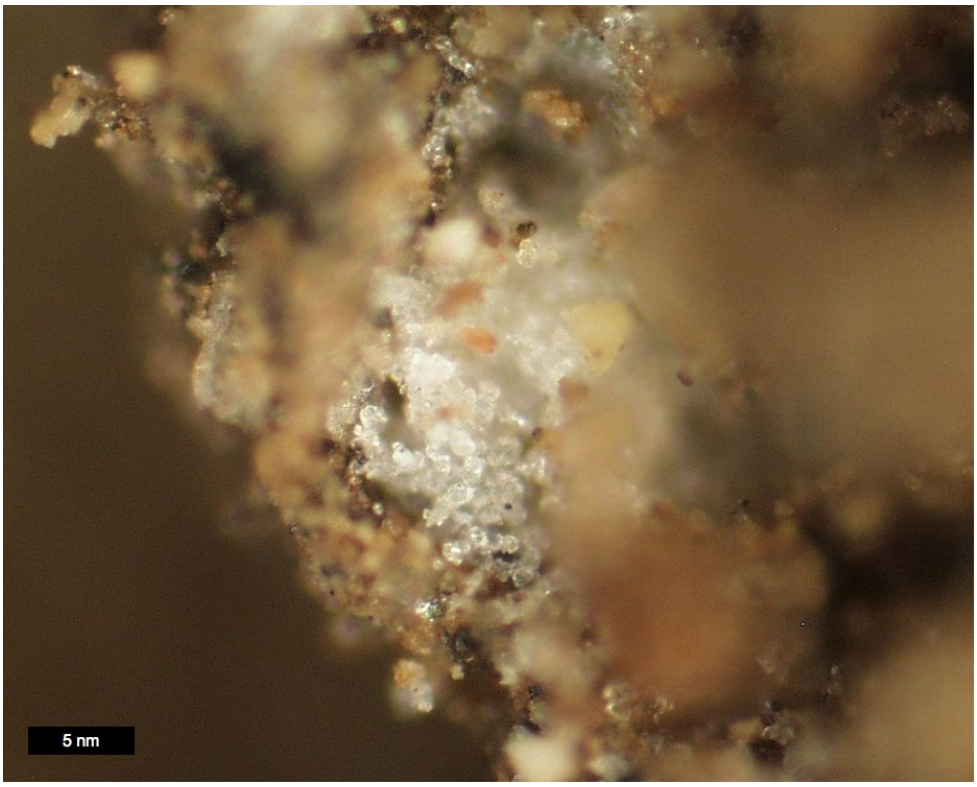


Figure B.129: Degraded woody plant tissue. Note that the granular deposit in the centre of the photo resembles damaged starch and ashy sediments. Magnification 200.

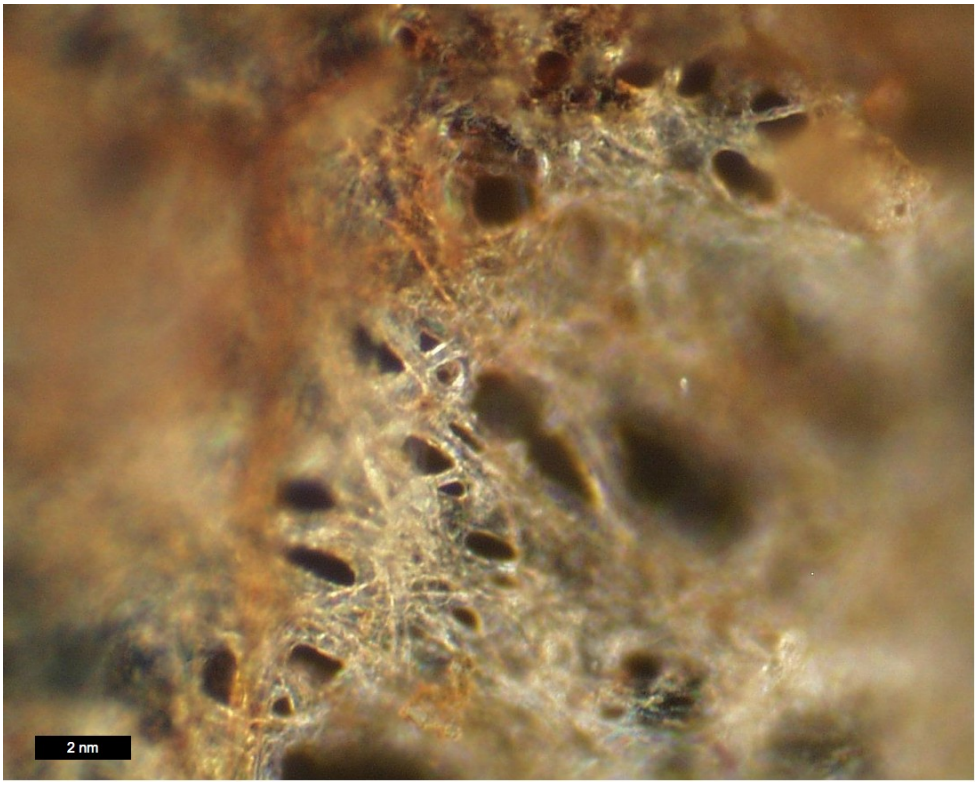


Figure B.130: Gelatinous hyphae. Thick hyphae deposits can be extremely similar to muscle tissue residues. The organic shapes and the holes are sometimes strikingly similar. Magnification 500, brightfield polarised light.

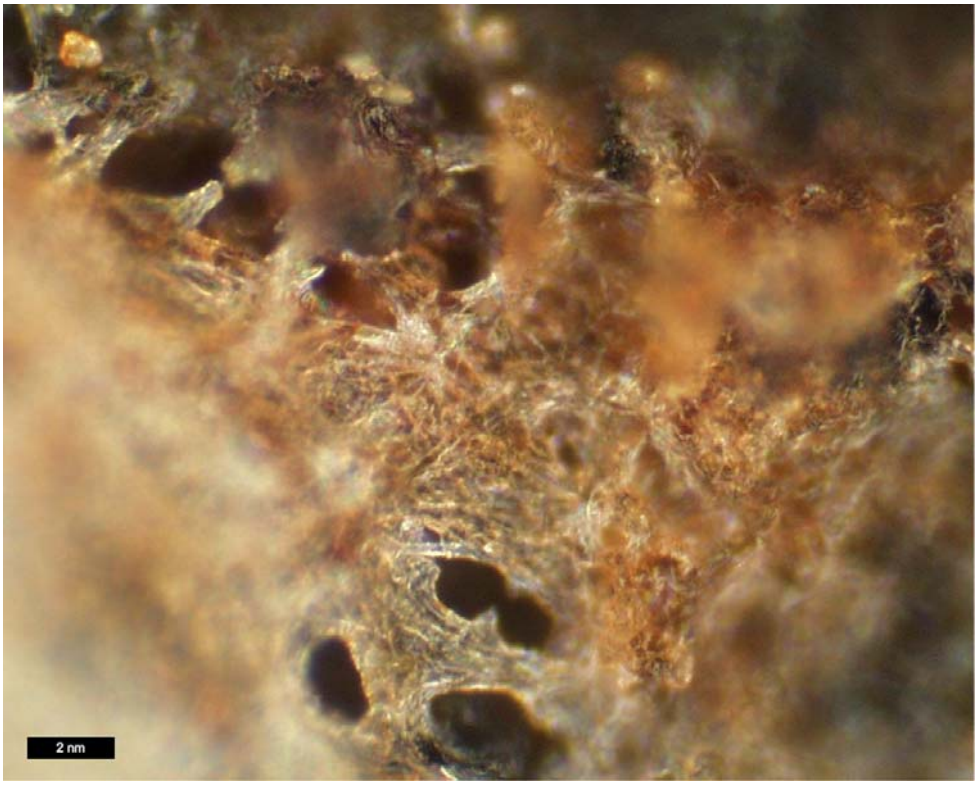


Figure B.131: Gelatinous hyphae. Thick hyphae deposits can be extremely similar to muscle tissue residues. The organic shapes and the holes are sometimes strikingly similar. Compare this photo to A.52. Magnification 500, brightfield polarised light.

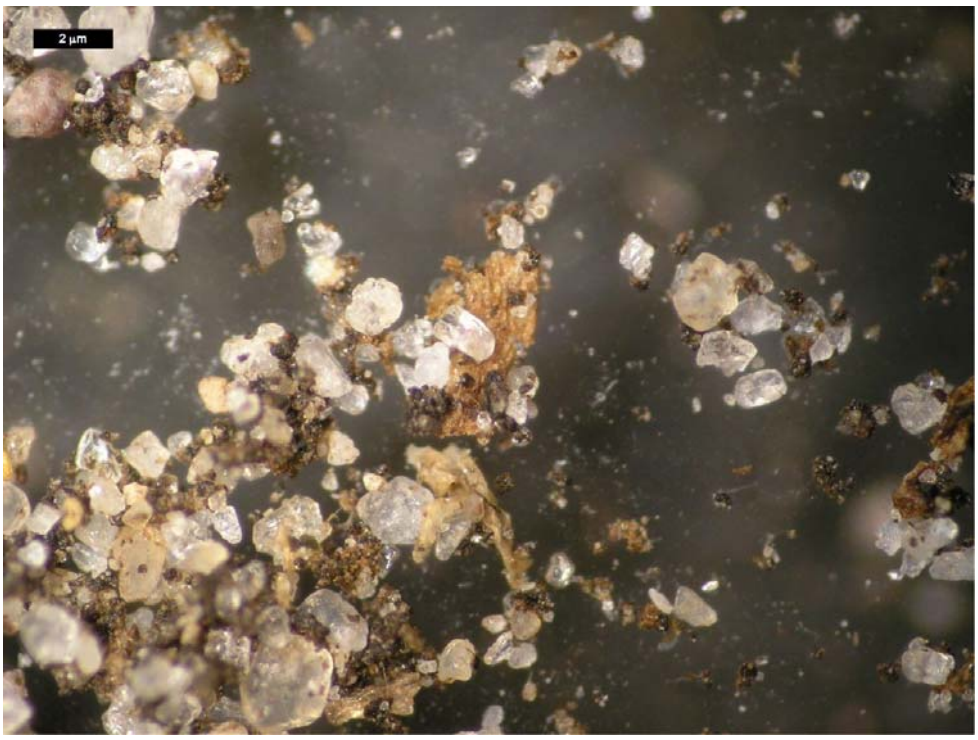


Figure B.132: Glass slide with sediment remains. Magnification 50.

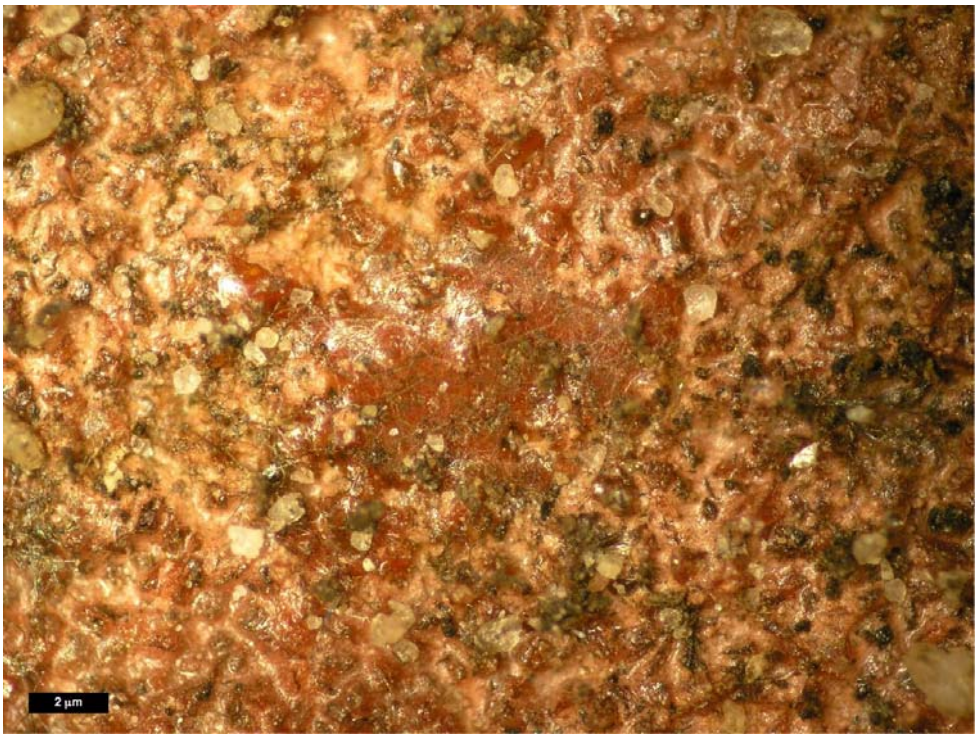


Figure B.133: Sandpaper surface "small". There are many smaller grains close to each other. Magnification 50.

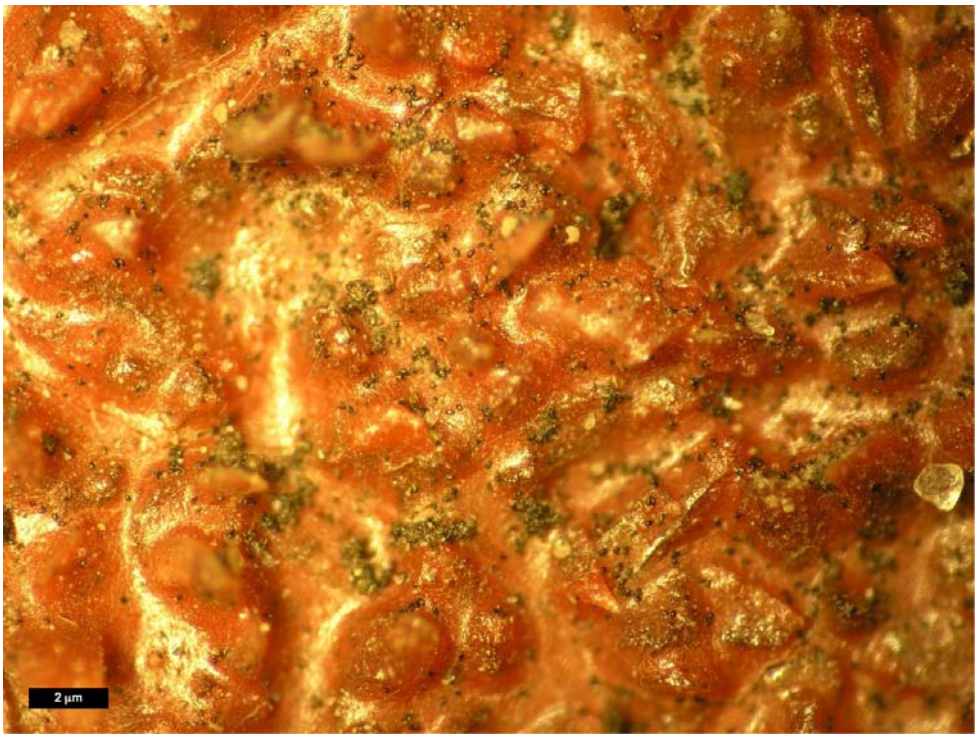


Figure B.134: Sandpaper surface "medium". Magnification 50.

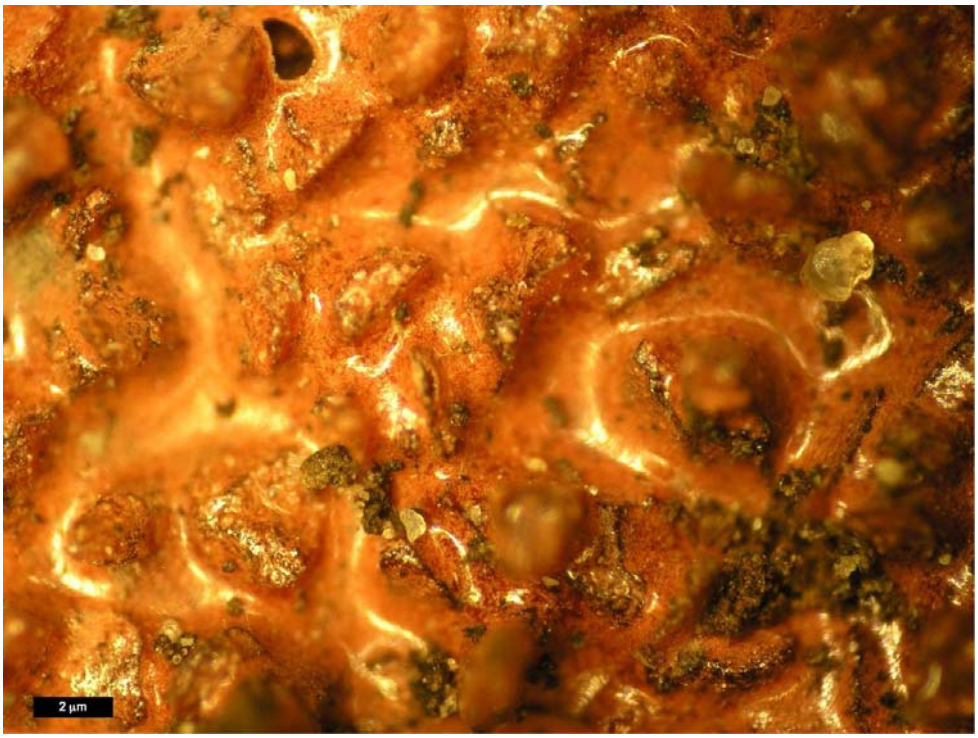


Figure B.135: Sandpaper surface “large”. There are fewer grains per cm2, compared to small or medium sized sandpaper. Magnification 50.

C APPENDIX: RESULTS FROM THE ANCIENT STERKFONTEIN SAMPLE

This appendix presents the results from Chapter Five: the Sterkfontein archaeological results. The appendix consists of micrographs and distribution maps on which the locations of all residues are indicated per stone tool. Unless stated otherwise all micrographs are taken in darkfield light using no filters. The key for the distribution maps is as follows:

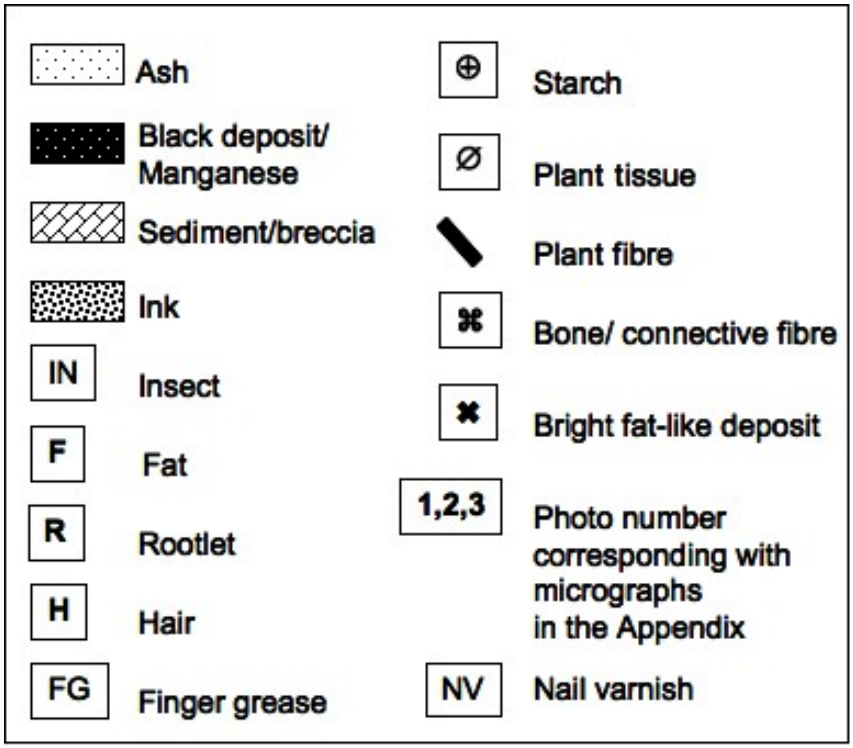


Figure C.1: Key to Figures.

Some maps have arrows and numbers, which indicate the location of micrographs. These micrographs are presented underneath the distribution map and start with the assigned map number. All the tools in the Figures are orientated with the distal end facing upwards.

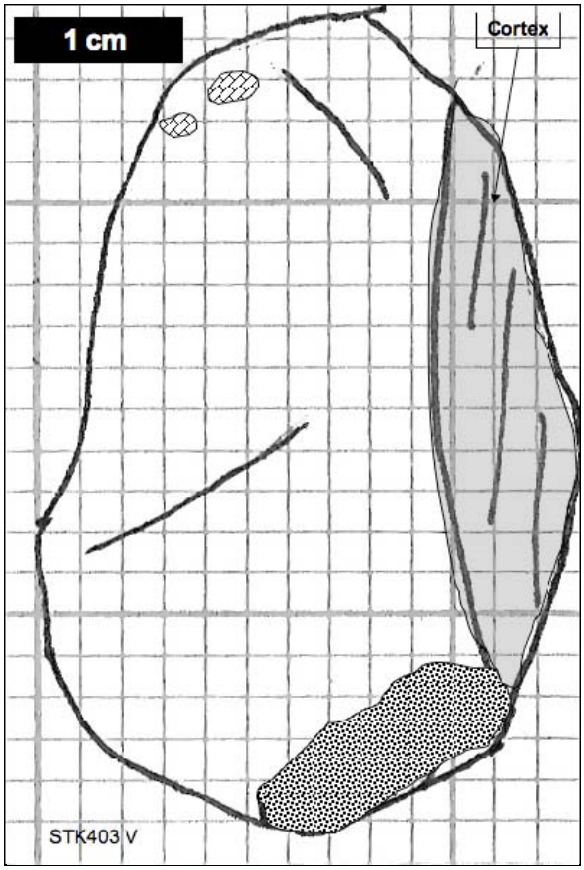


Figure C.2: Distribution map for STK403, ventral side. No organic remains are present. For key see Figure C.1.

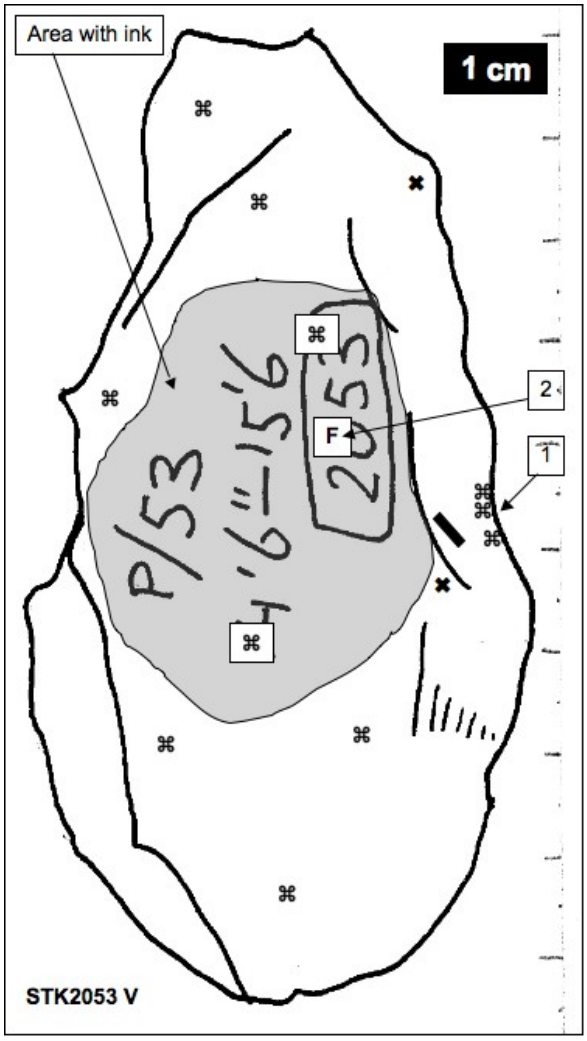


Figure C.3: Distribution map for STK2053, ventral side. Note the large areas with writing and ink. In this ink area and also on top of the label bone and fat are present. For key see Figure C.1.

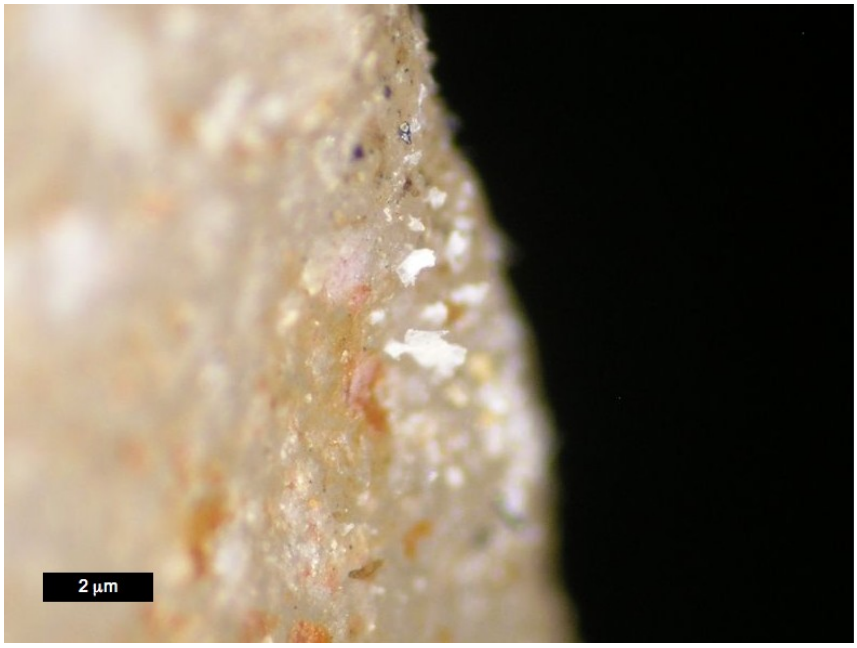


Figure C.4: 1= Micrograph of white bone-like residues on STK2053. The orange spots are sediment. Magnification 50.

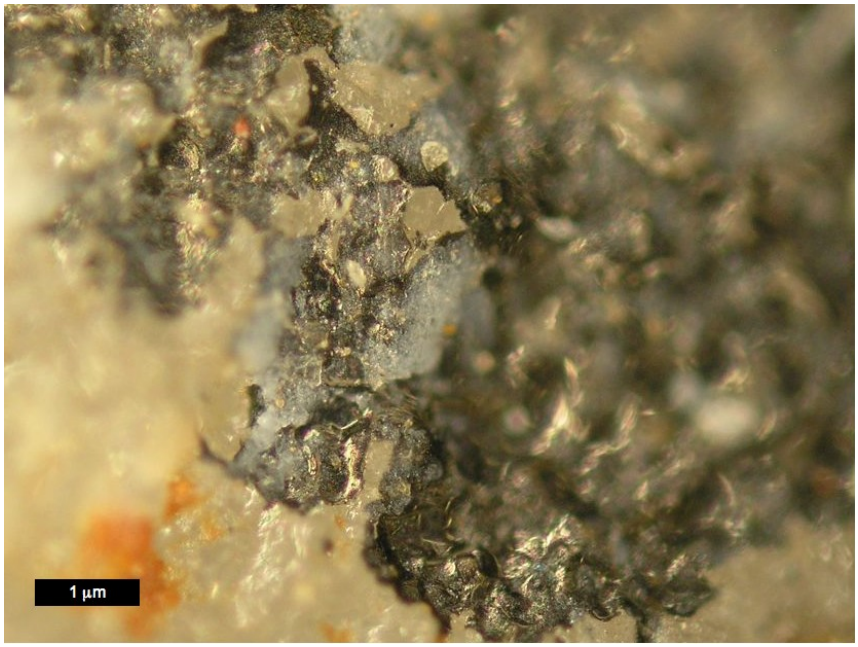


Figure C.5: 2= Micrograph of white fat on ink, STK2053. Magnification 100.

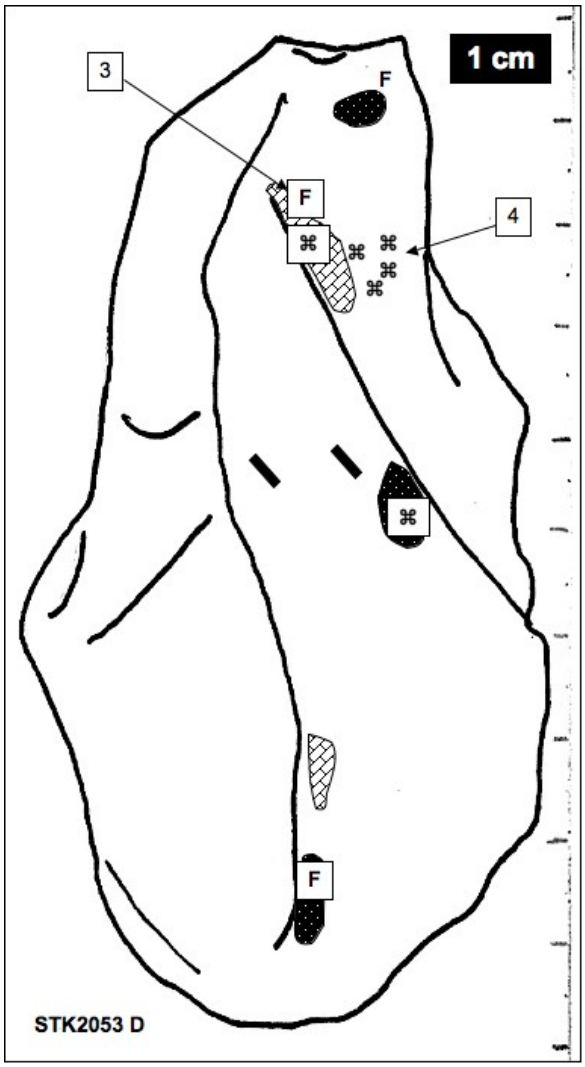


Figure C.6: Distribution map for STK2053, dorsal side. Note the fat and collagen deposits on sediment and manganese stains. For key see Figure C.1.

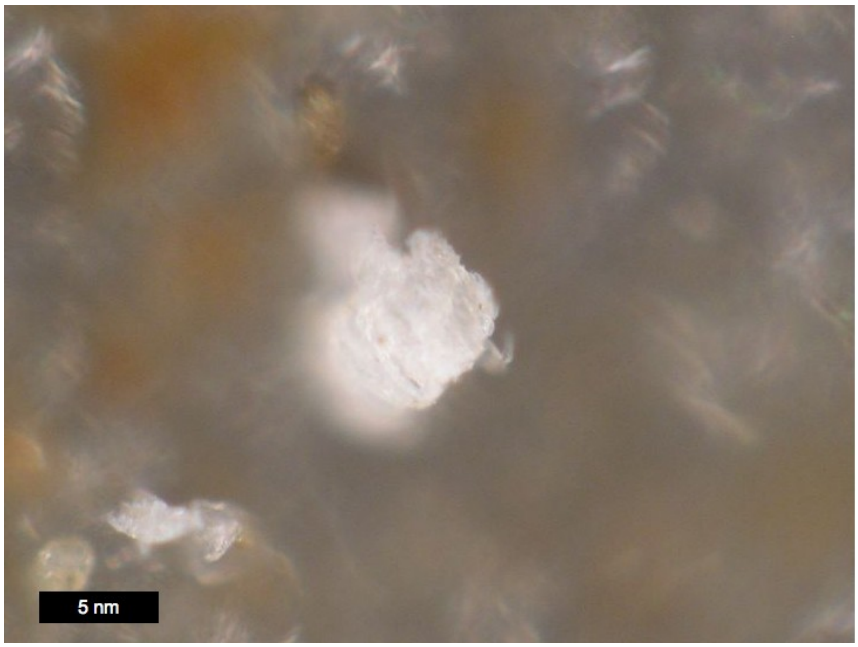


Figure C.7: 3= Micrograph of fat on STK2053. The orange spots in the background are sediment. Magnification 200.



Figure C.8: 4= Micrograph of fat on STK2053. The white fat deposit lays on top of the orange sediment. Magnification 100.

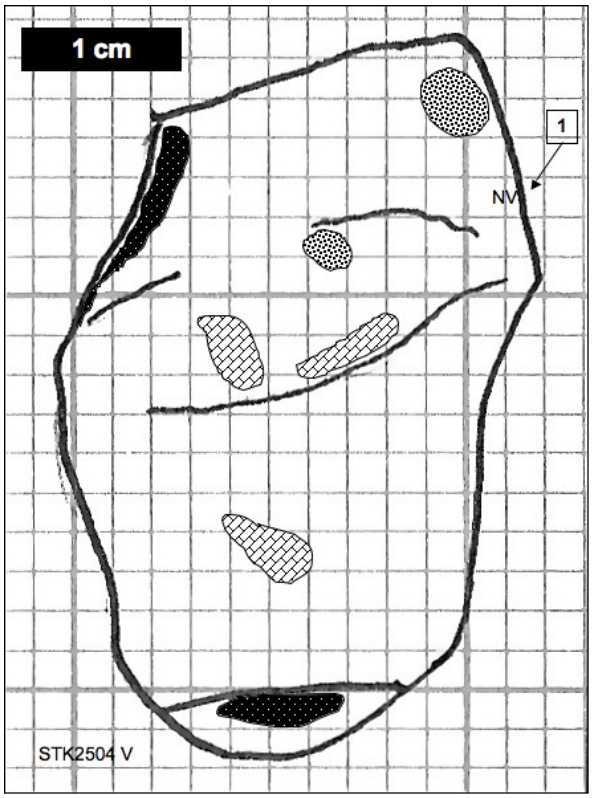


Figure C.9: Distribution map for STK2504, ventral side. For key see Figure C.1.



Figure C.10: 1= Micrograph of nail varnish on the edge of STK2504; note the air bubbles indicated with the arrow. Magnification 50.

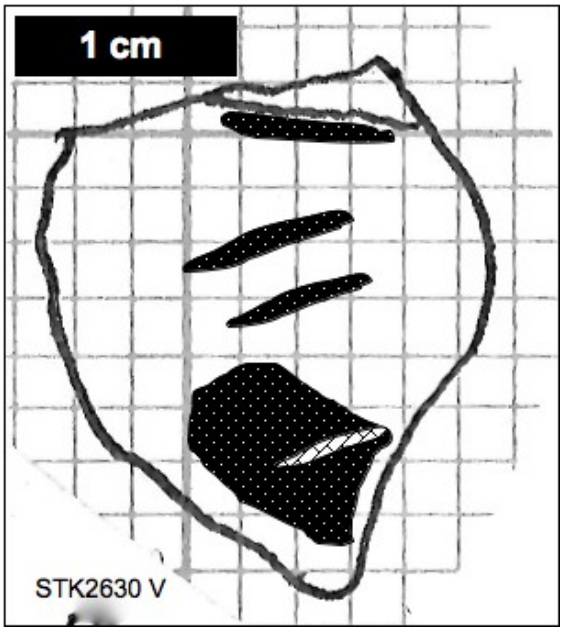


Figure C.11: Distribution map of STK2630, ventral side. Note the large black manganese stains. For key see Figure C.1.

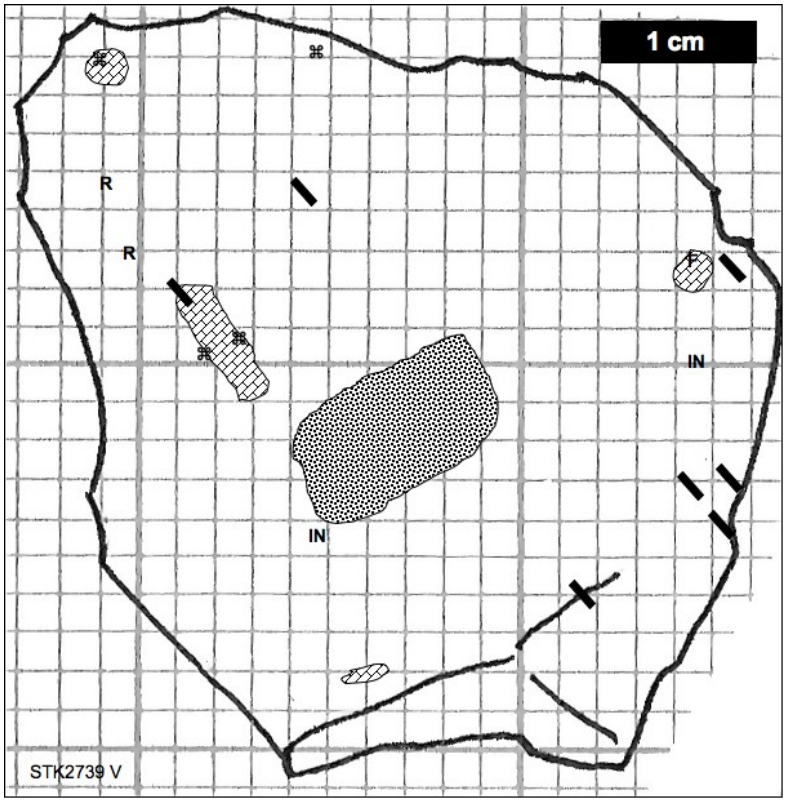


Figure C.12: Distribution of STK2739, ventral side. The plant fibres are spread randomly over the surface. Note the bone deposits on top of the sediment deposits. For key see Figure C.1.

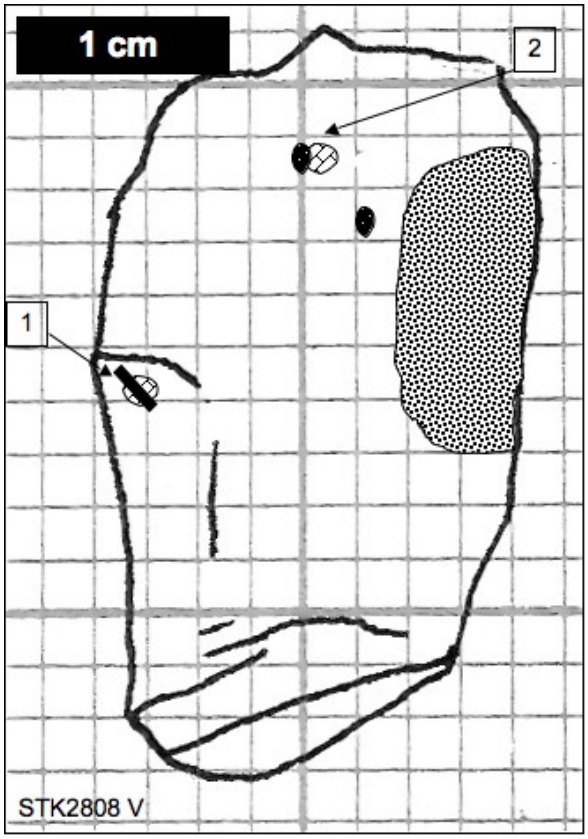


Figure C. C.13: Distribution map for STK2808, ventral side. For key see Figure C.1.



Figure C.14: 1= Micrograph of a plant fibre on top of the orange sediment on STK2808. Magnification 100.

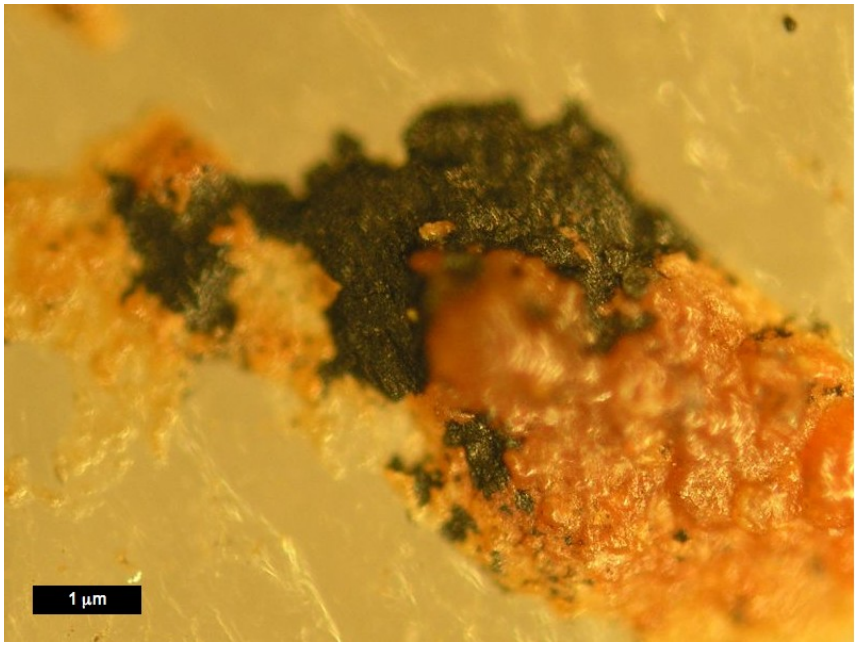


Figure C.15: 2= Micrograph of a black manganese and a sediment deposit on STK2808. Magnification 100.

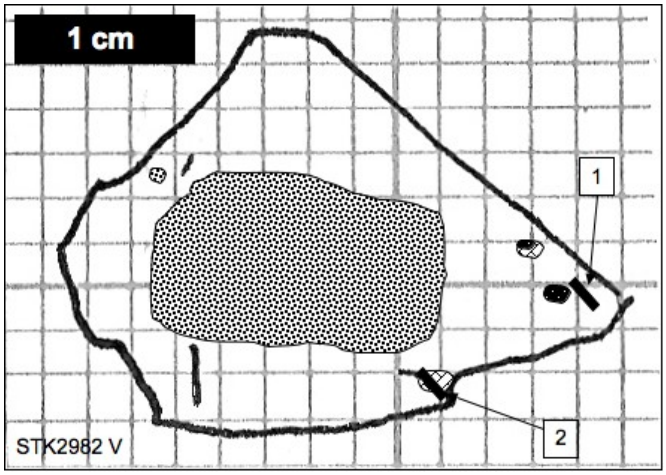


Figure C.16: Distribution map for STK2982, ventral side. Note the tool's label, which covers a large part of the surface. For key see Figure C.1.

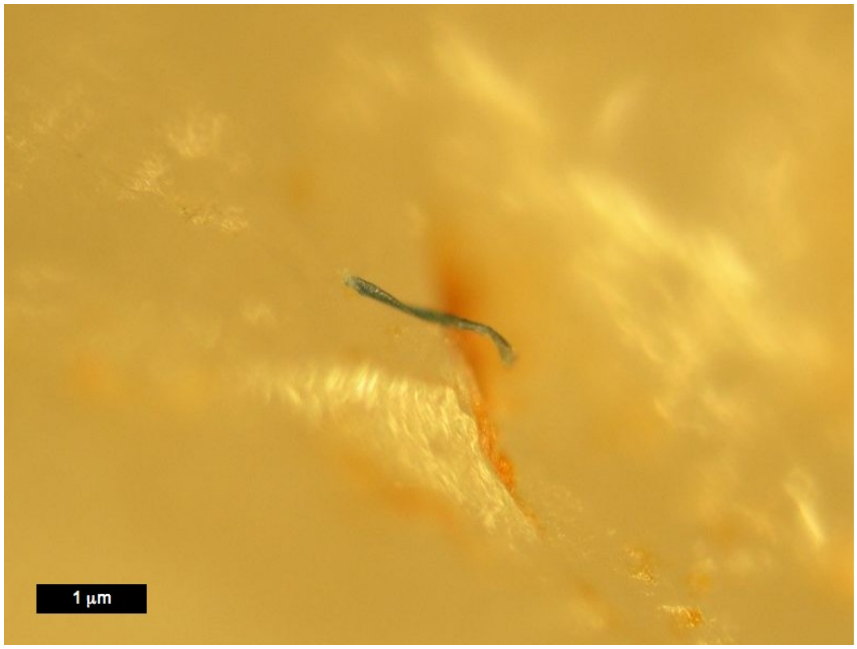


Figure C.17: 1= Micrograph of a blue fibre on STK2982. Magnification 100.



Figure C.18: 2= Micrograph of a plant fibre on sediment on STK2982. Magnification 50.

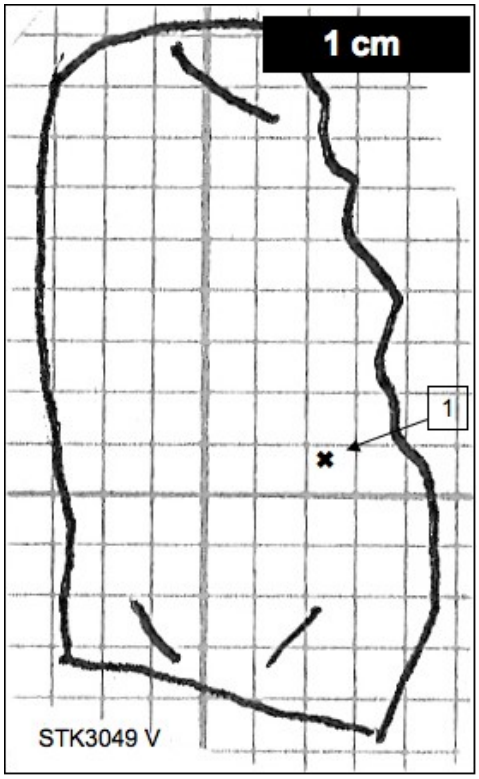


Figure C.19: Distribution map for STK3049, ventral side. For key see Figure C.1.

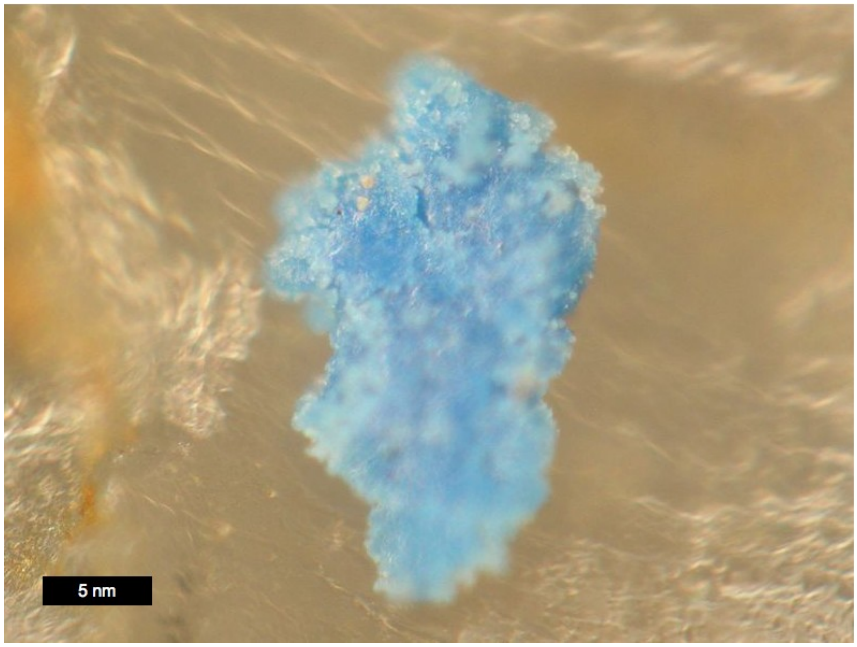


Figure C.20: 1= Micrograph of blue fat-like deposit. The bright colour indicates that this fat-like residue is a contaminant. Magnification 200.

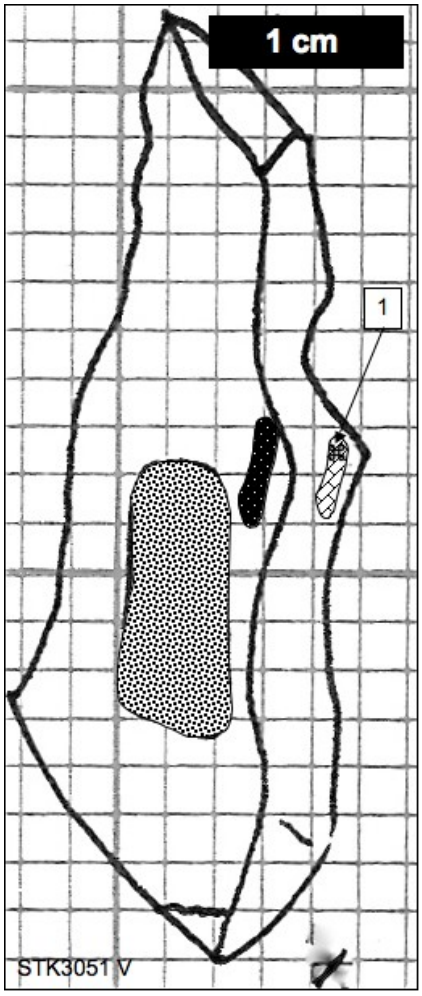


Figure C.21: Distribution map for STK3051, ventral side. For key see Figure C.1.

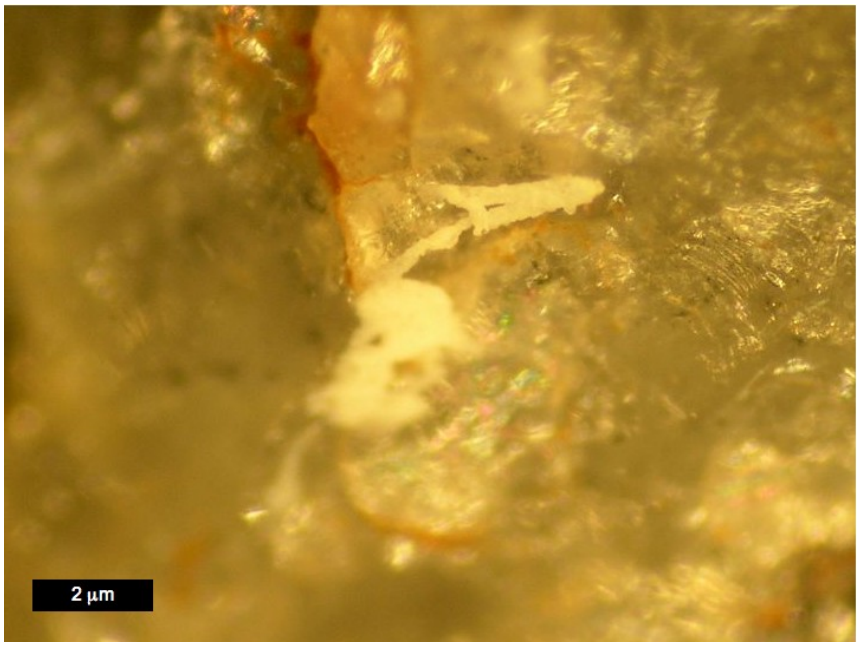


Figure C.22: 1= Micrograph of a connective fibre on STK3051. The fibre lays on top of a light sediment layer. Magnification 50.

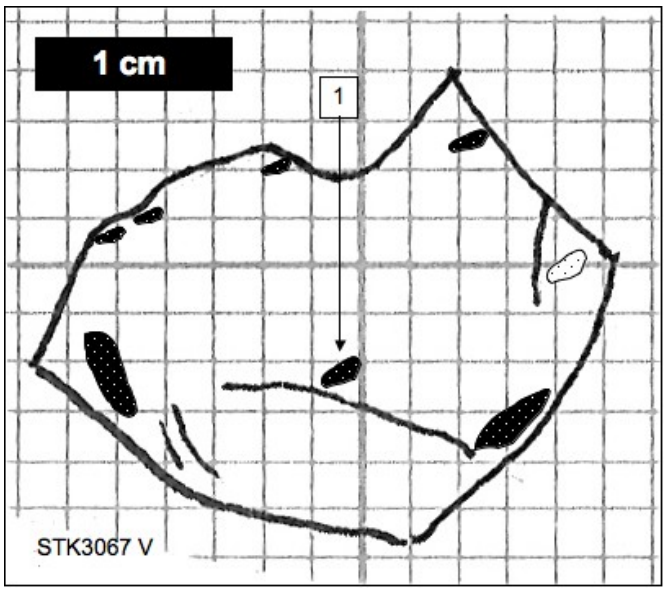


Figure C.23: Distribution map for STK3067, ventral side. Note that black manganese stains are abundant. For key see Figure C.1.

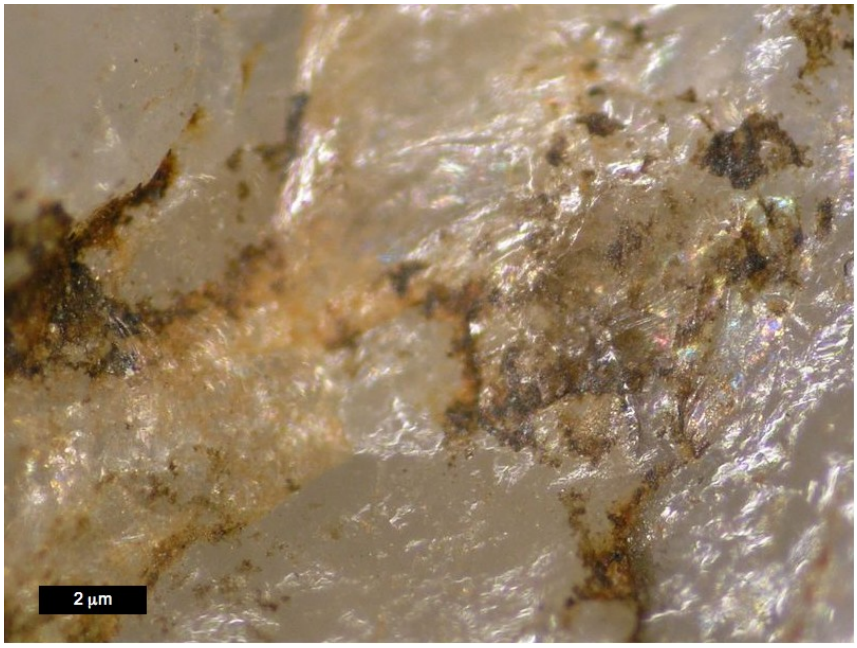


Figure C.24: 1= Micrograph of a manganese stain on STK3067. Magnification 50.

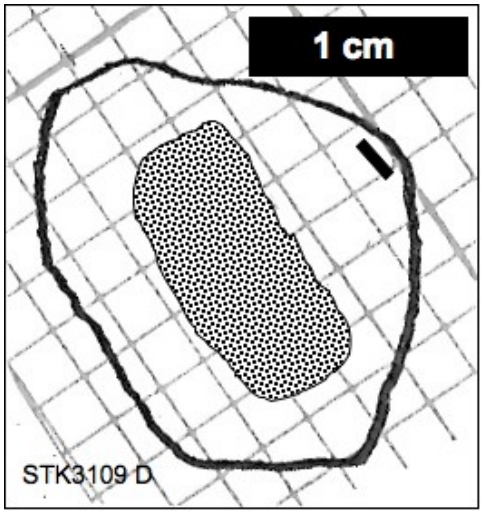


Figure C.25: Distribution map for STK3109, dorsal side. A large part of this flake is covered with the tool's label. For key see Figure C.1.

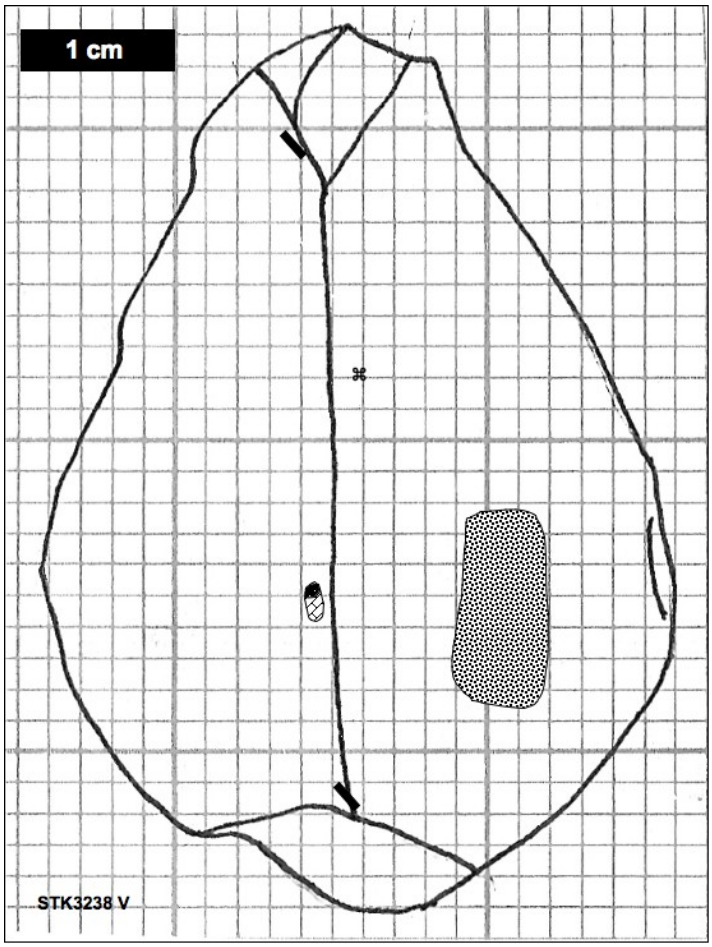


Figure C.26: Distribution map for STK3238, ventral side. For key see Figure C.1.

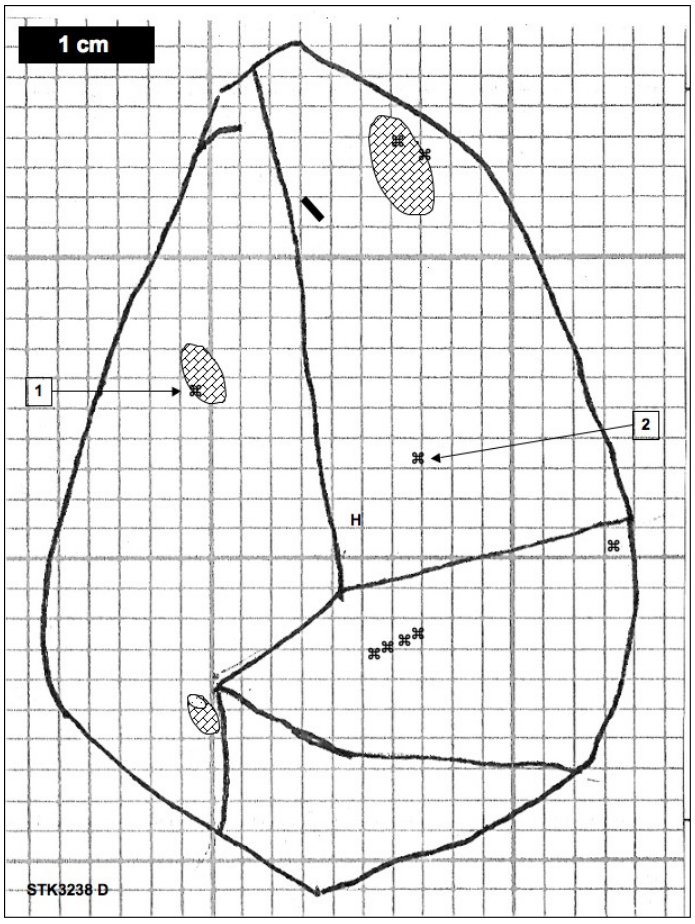


Figure C.27: Distribution map for STK3238, dorsal side. Because fatty bone is also present on top of sediment patches, it is unlikely that the other fatty bone deposits are use-related. For key see Figure C.1.



Figure C.28: 1= Micrograph of fatty bone on STK3238. The residue is clearly deposited on top of sediment. Magnification 50.

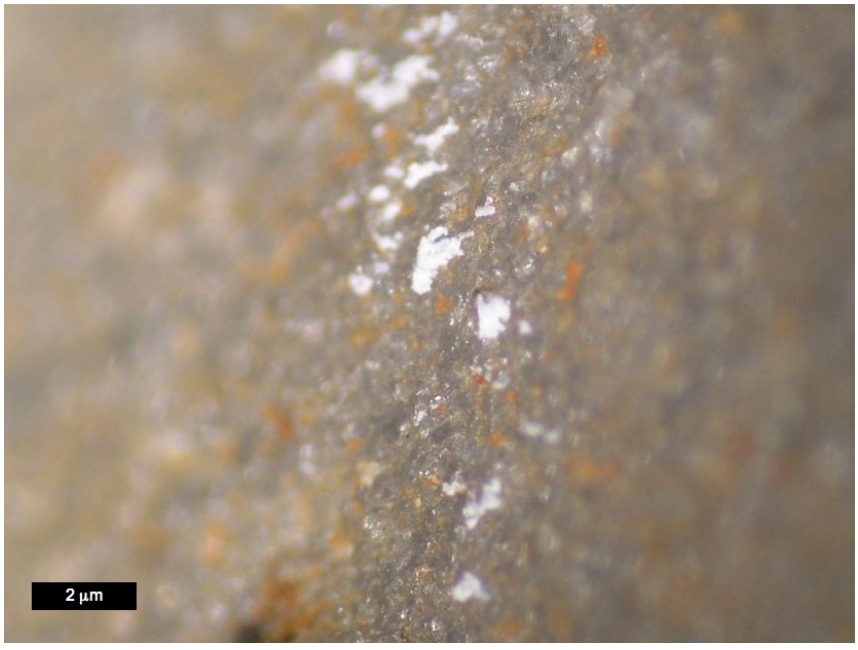


Figure C.29: 2= Micrograph of fatty bone on STK3238. Note that the bone is deposited lightly and appears to "float" over the surface. It is not smeared. Magnification 50.

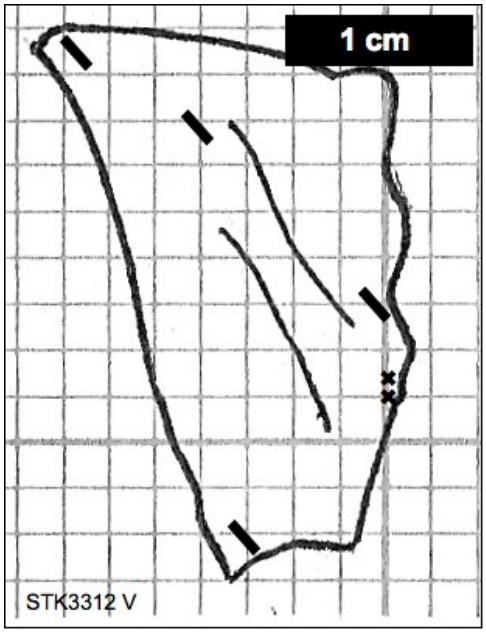


Figure C.30: Distribution map for STK3312, ventral side. For key see Figure C.1.

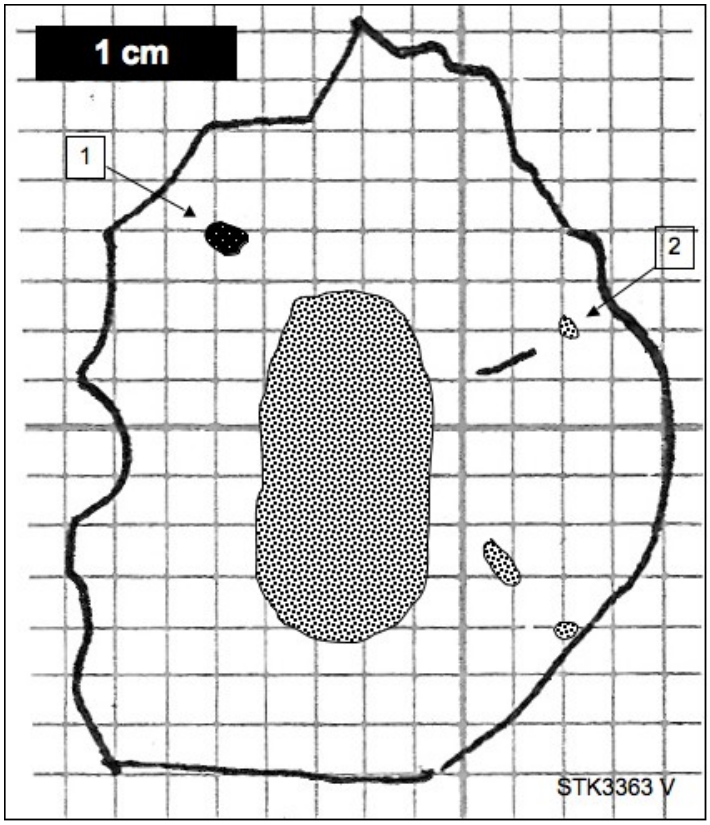


Figure C.31: Distribution map for STK3363, ventral side. For key see Figure C.1.

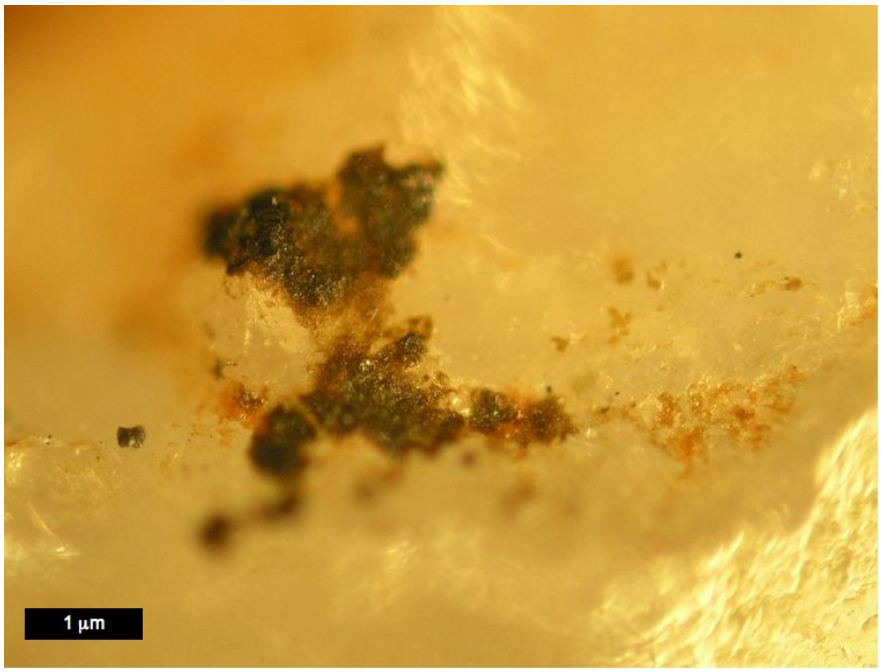


Figure C.32: 1= Micrograph of a browner manganese deposit on STK3363. Magnification 100.

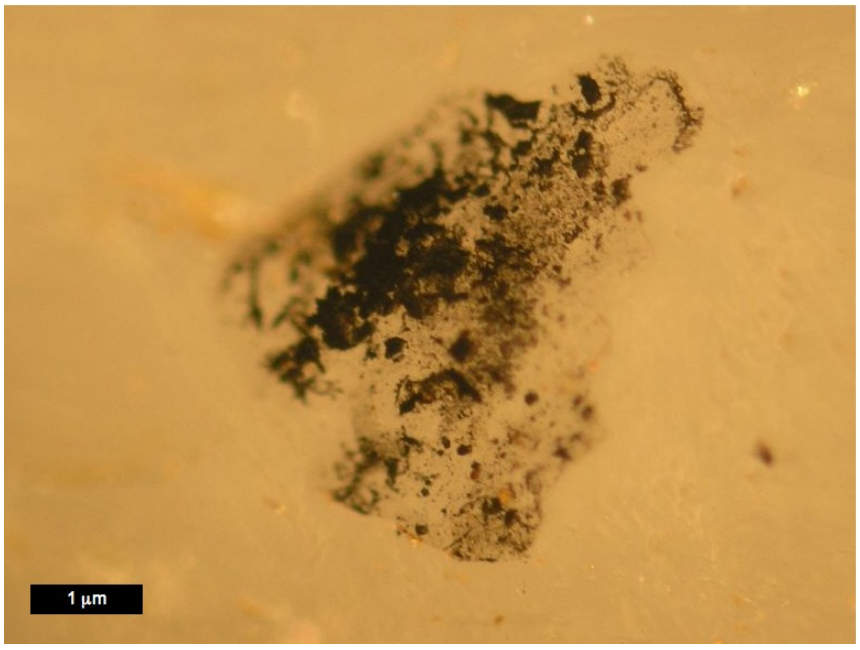


Figure C.33: 2= Micrograph of an ink deposit on STK3363. Magnification 100, brightfield polarised light.

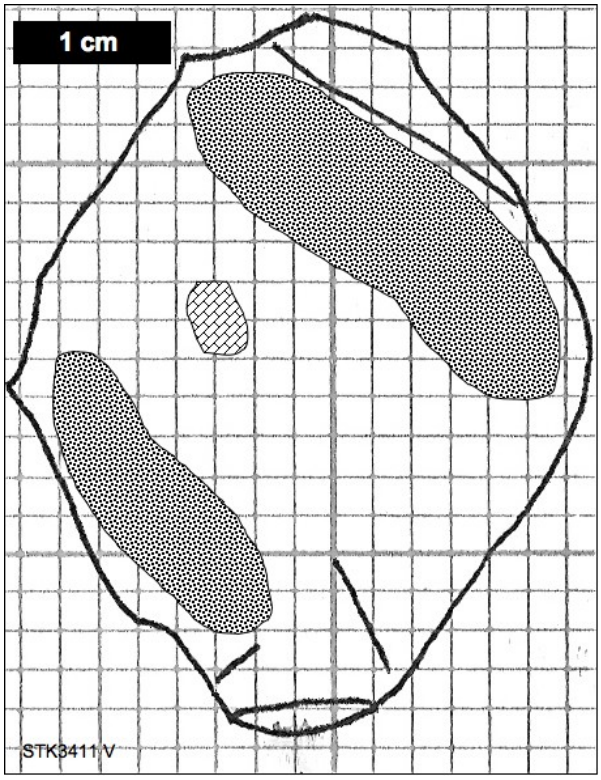


Figure C.34: Distribution map for STK3411, ventral side. Note the areas with writing. For key see Figure C.1.

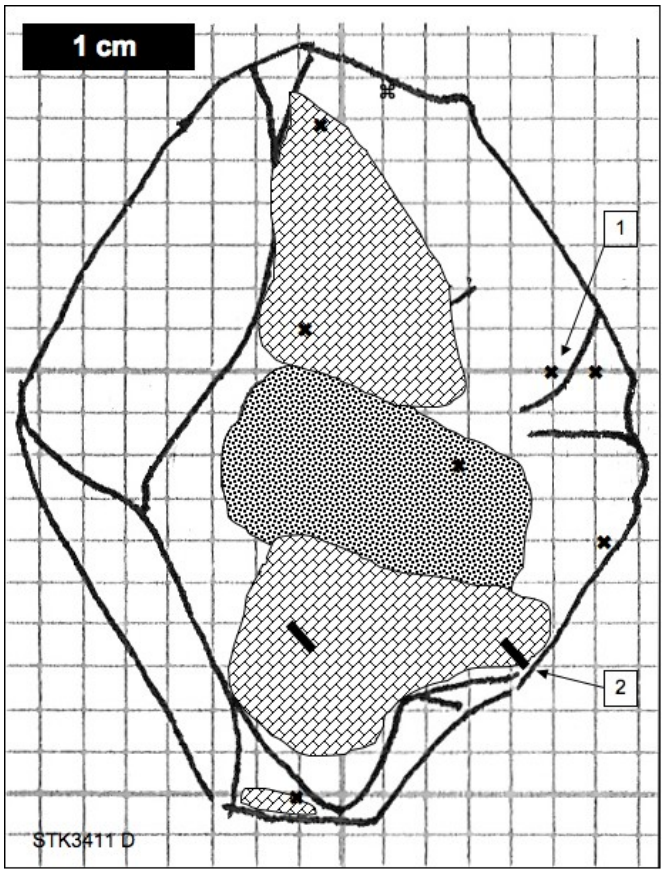


Figure C.35: Distribution map for STK3411, dorsal side. For key see Figure C.1.

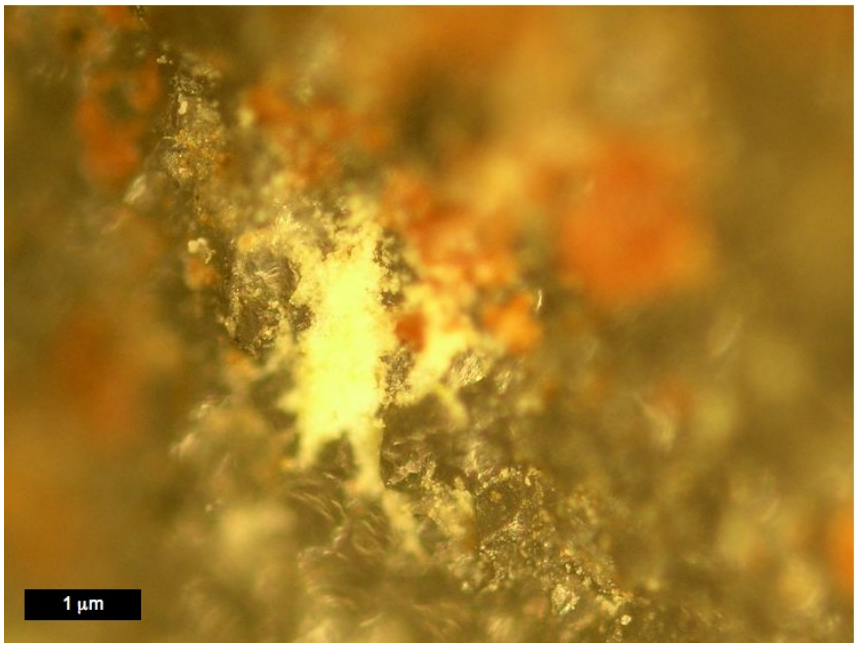


Figure C.36: 1= Micrograph of light green fat-like deposit on STK3411. Magnification 100.

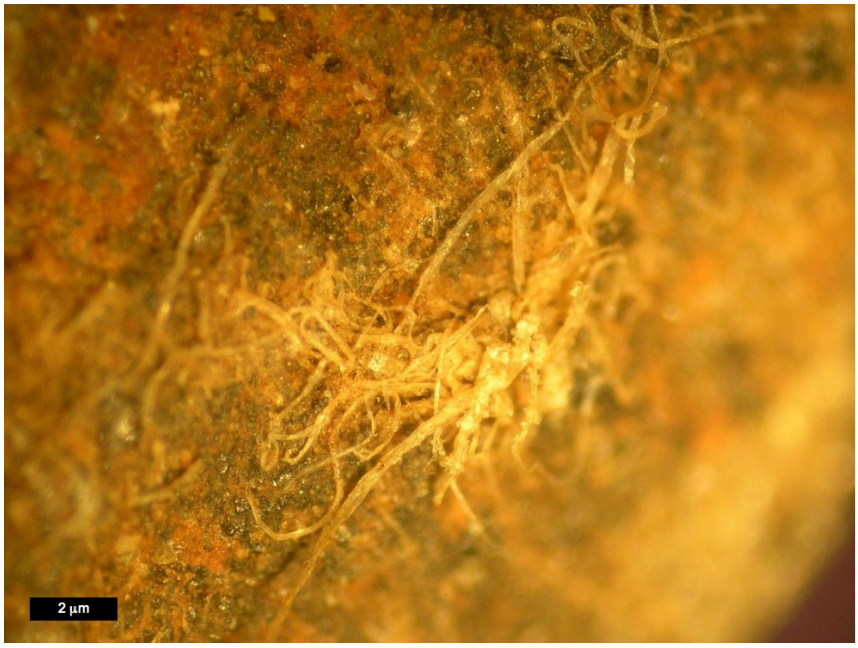


Figure C.37: 2= Micrograph of plant fibres on sediment on STK3411. Magnification 50.

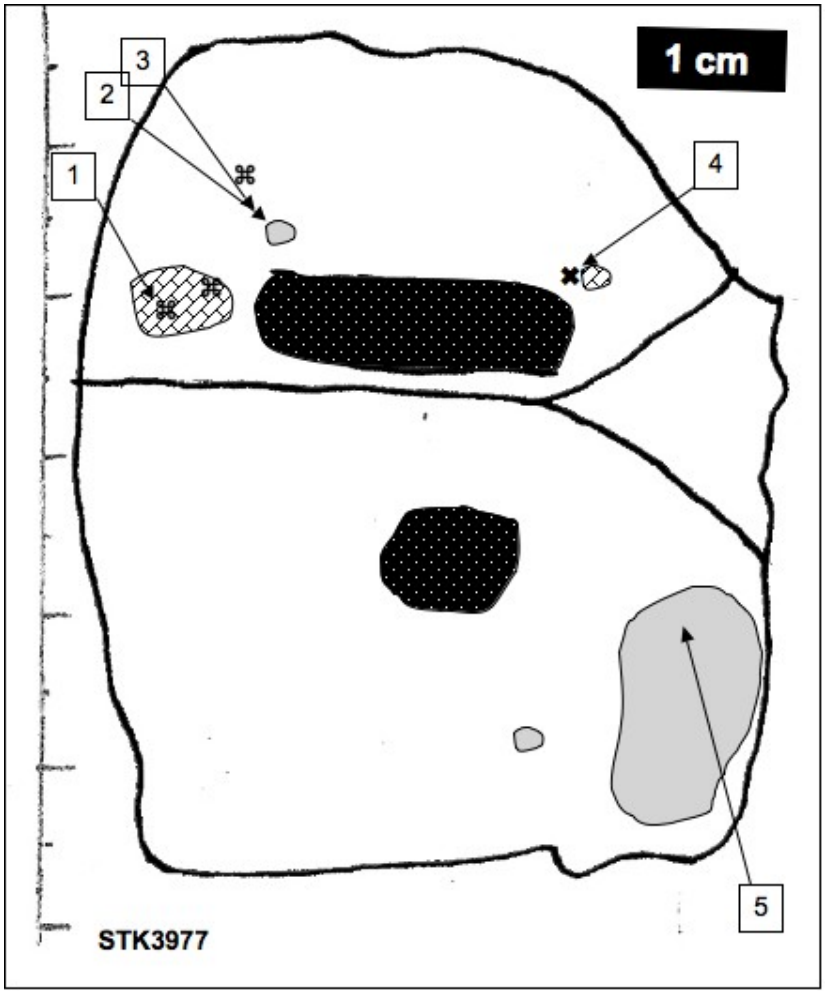


Figure C.38: Distribution map for STK3977. For key see Figure C.1.

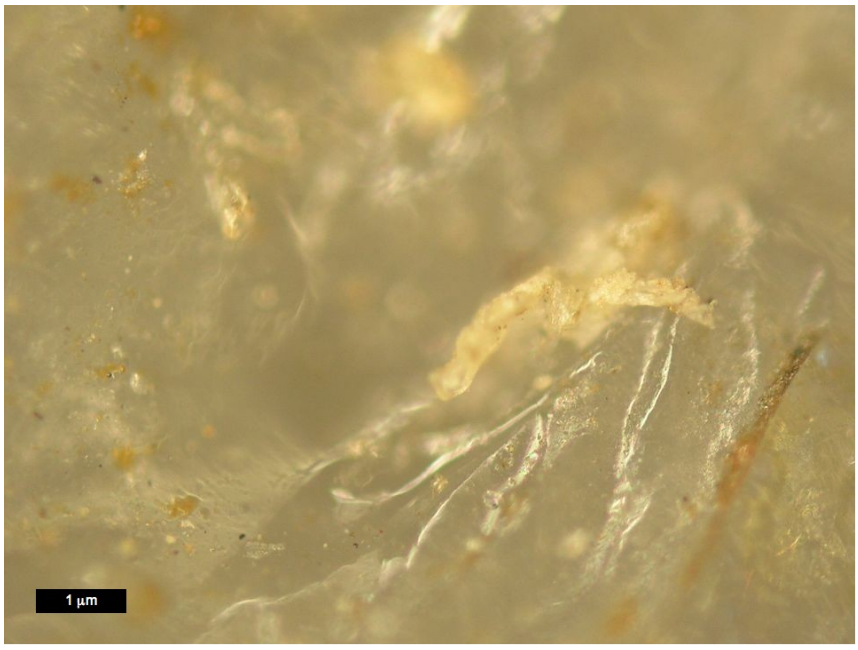


Figure C.39: 1= Micrograph of connective fibrous material on STK3977. Magnification 100.

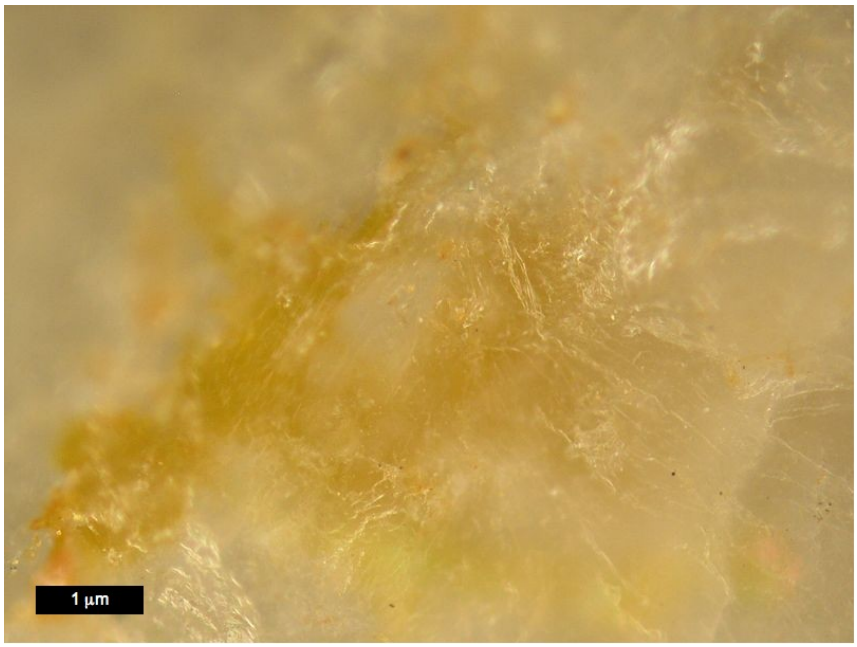


Figure C.40: 2= Micrograph of yellow inclusion in STK3977, which could be confused for a stain of a liquid deposit. Magnification 100.

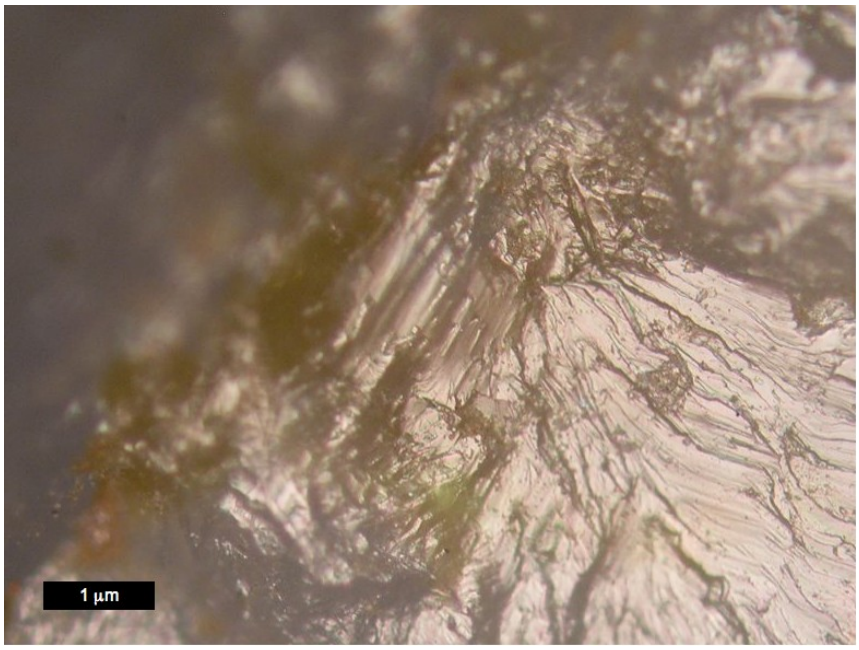


Figure C.41: 3= Micrograph of yellow inclusion in STK3977. Magnification 100, brightfield cross-polarised light.

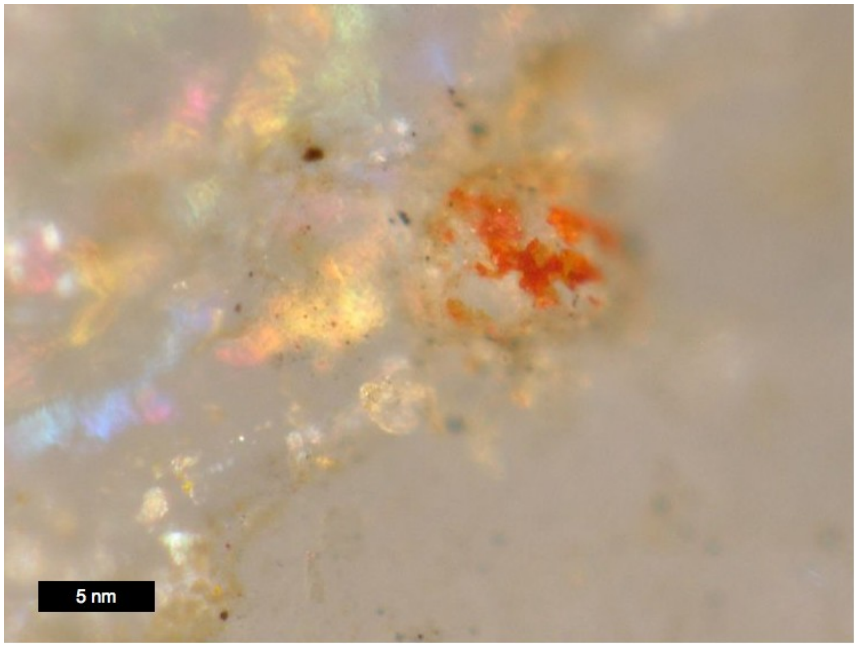


Figure C.42: 4= Micrograph of red fat-like deposit on STK3977. Magnification 200, brightfield polarised light.

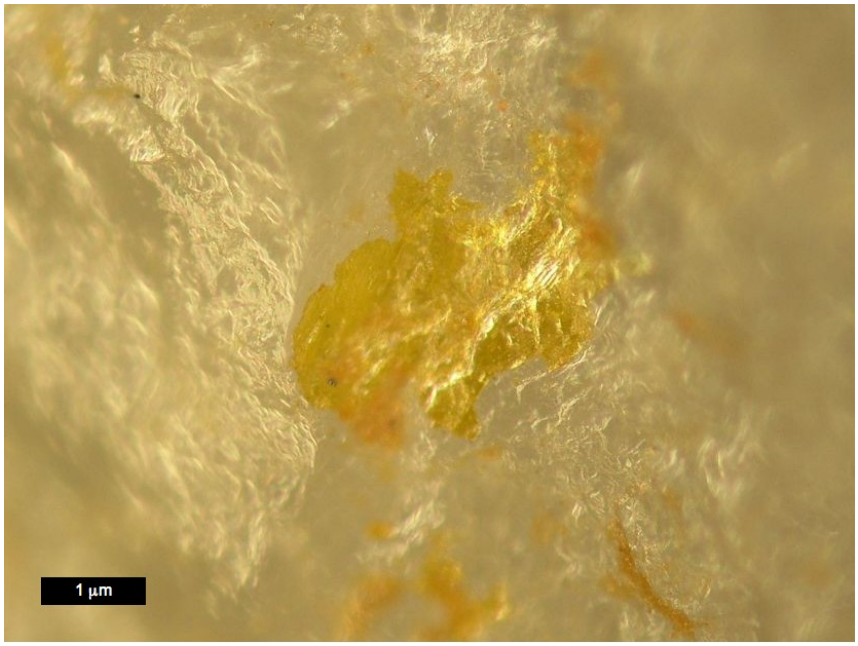


Figure C.43: 5= Micrograph of yellow inclusion in STK3977. This inclusion is similar to the one in C.40 and C.41 and could easily be confused for a thin deposit. Magnification 100.

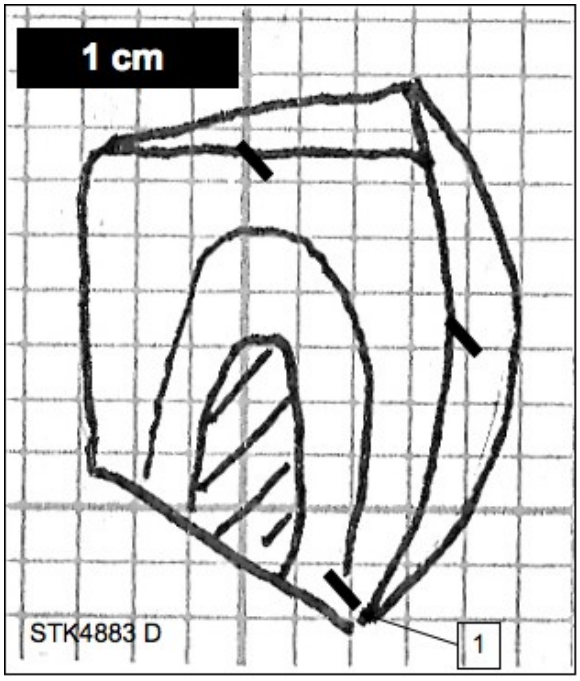


Figure C.44: Distribution map for STK4883, dorsal side. For key see Figure C.1.



Figure C.45: 1= Micrograph of plant fibres on STK4883. The plant fibres appear fresh and lay on top of orange sediment, indicating that the fibres are contaminants. Magnification 100.

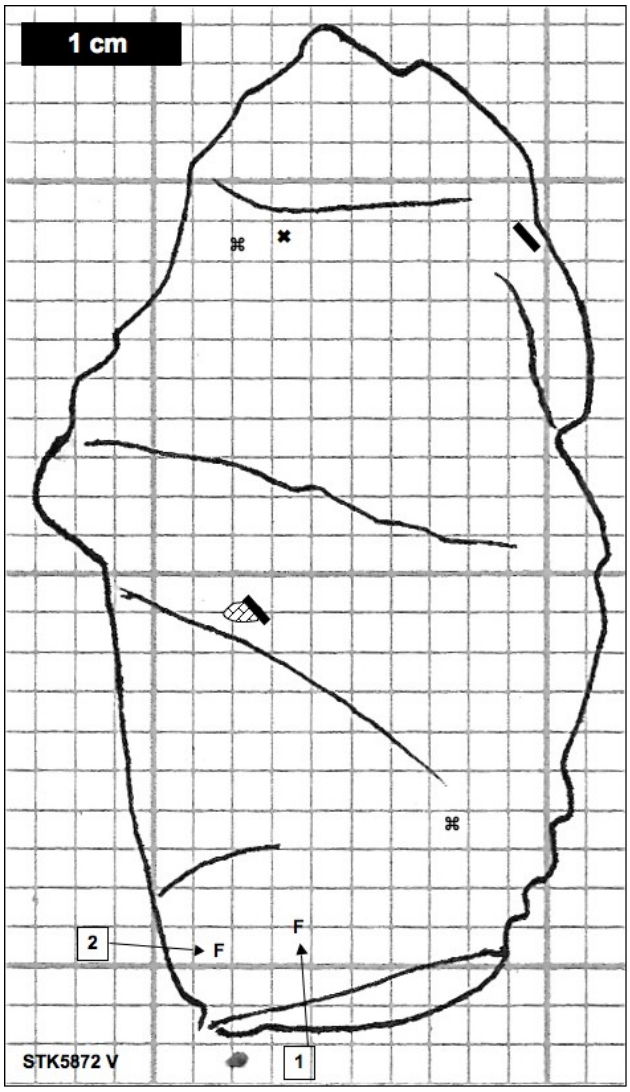


Figure C.46: Distribution map for STK5872, ventral side. For key see Figure C.1.

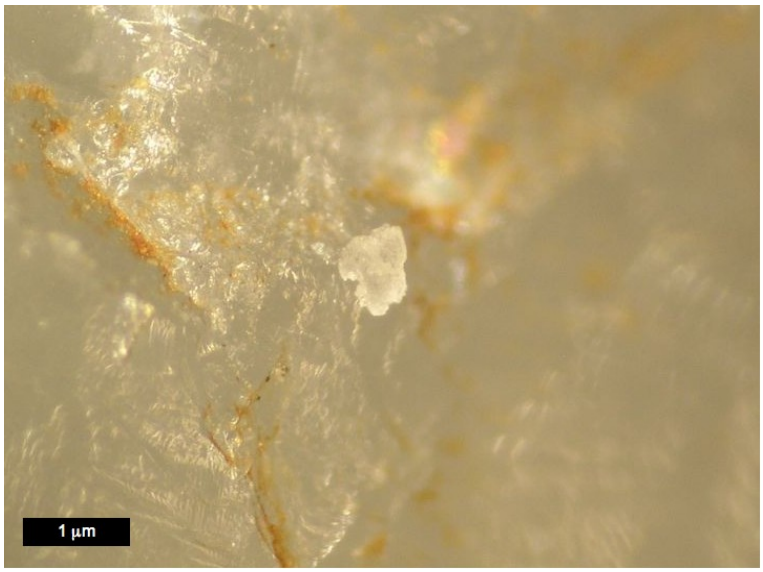


Figure C.47: 1= Micrograph of fat on STK5872. Note that the fat drop almost appears to "float" over the cracks with sediment. Magnification 100.

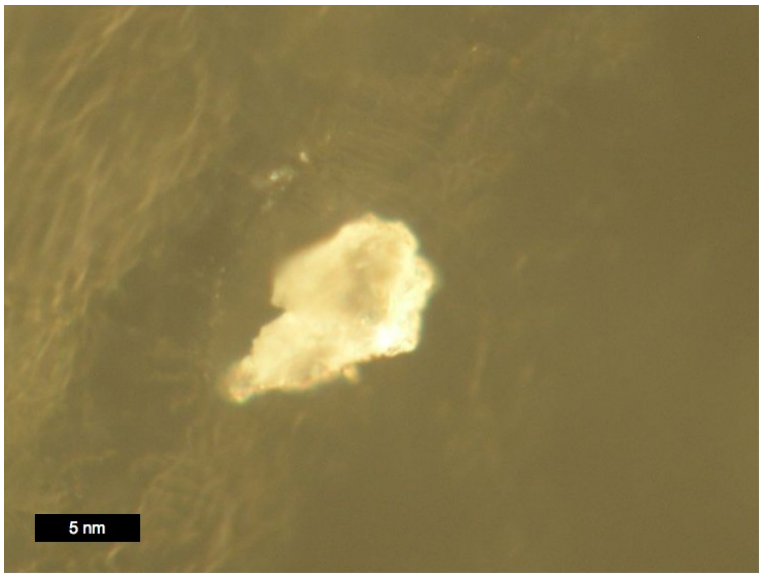


Figure C.48: 2= Micrograph of fat on STK5872. The fat deposit lays lightly over the surface. Use-related residues are generally smeared over the surface. Magnification 200, brightfield polarised light.

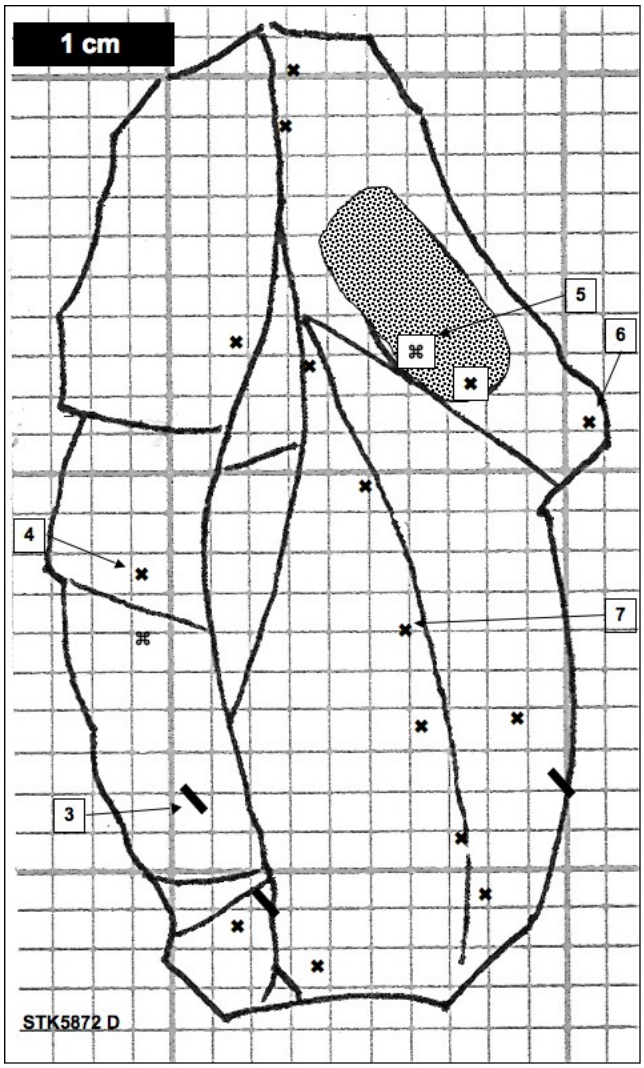


Figure C.49: Distribution map for STK5872, dorsal side. The large number of bright fat-like deposits indicates that this artefact is contaminated and that the other remains must be analysed with care. For key see Figure C.1.

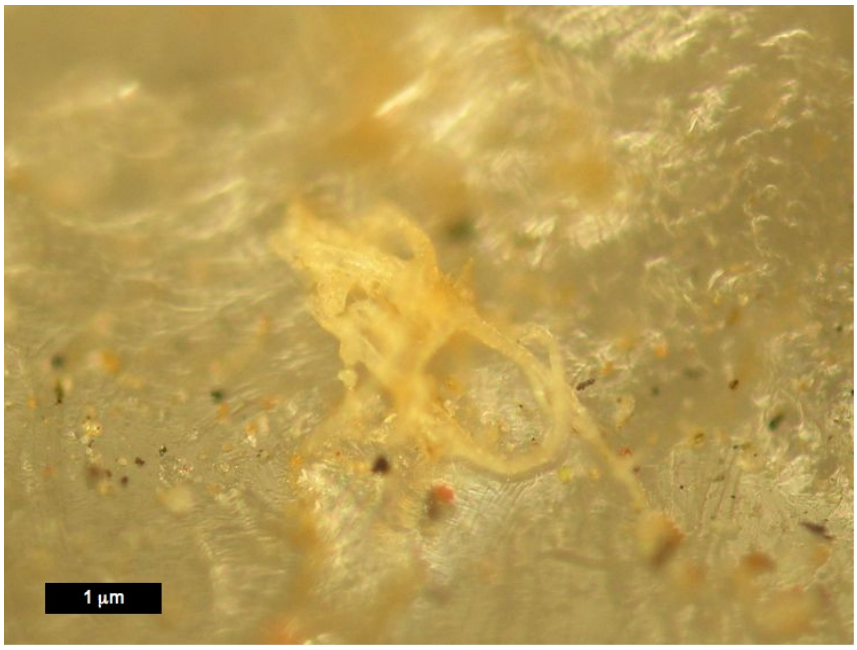


Figure C.50: 3= Micrograph of a fibre on STK5872. Magnification 100.

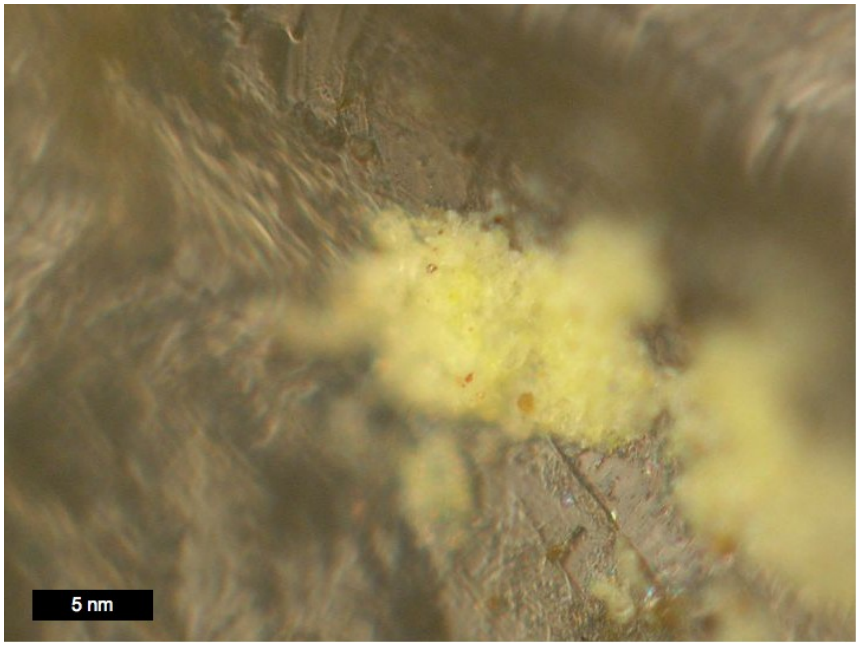


Figure C.51: 4= Micrograph of a green fat-like deposit on STK5872. Magnification 200, brightfield polarised light.

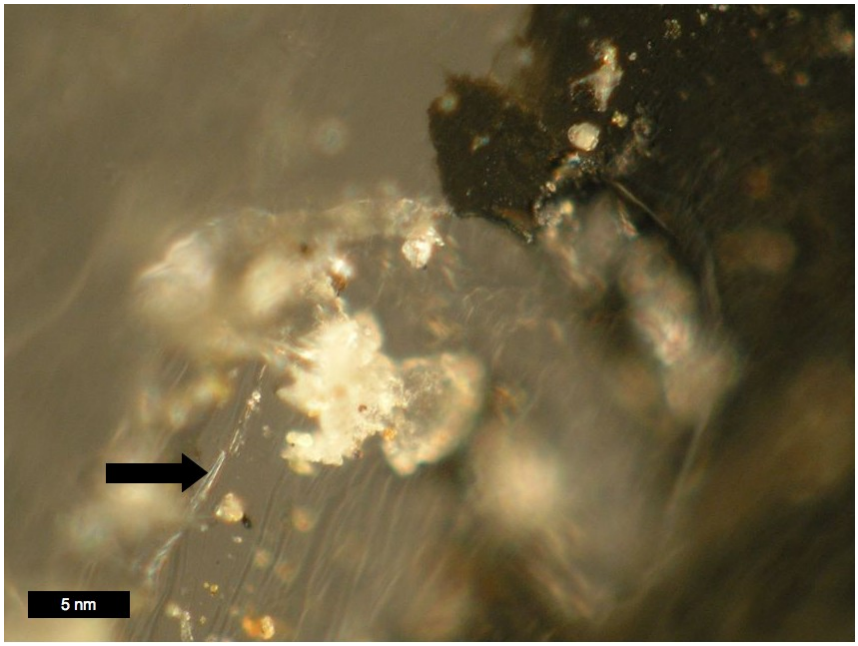


Figure C.52: 5= Micrograph of fatty bone on STK5872. This fatty bone is deposited on top of artefact's label. The arrow indicated the nail varnish; the black deposit is ink. Magnification 200.

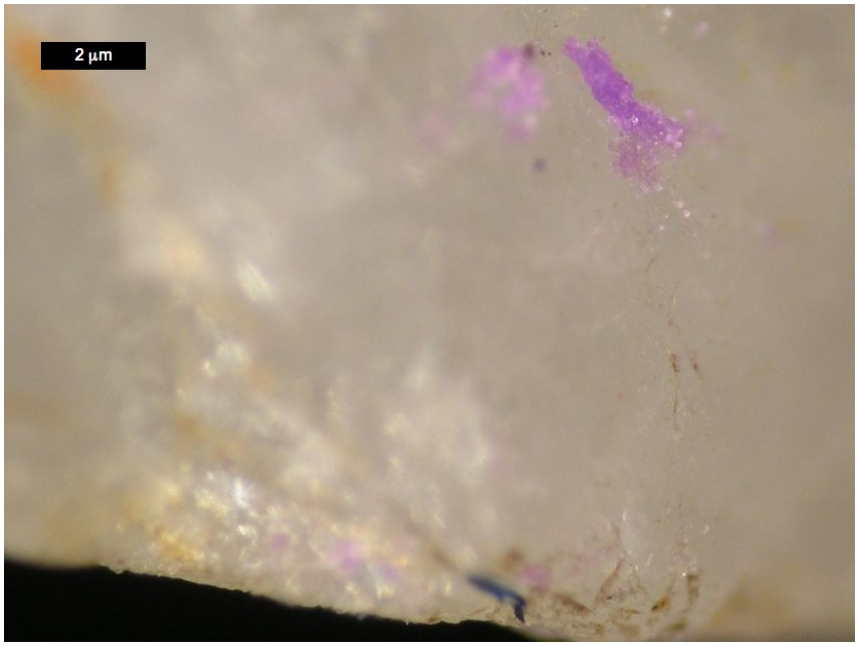


Figure C.53: 6= Micrograph of a purple fat-like deposit on STK5872; this is a clear contaminant. Note the blue modern fibre at the artefact's edge. Magnification 50.

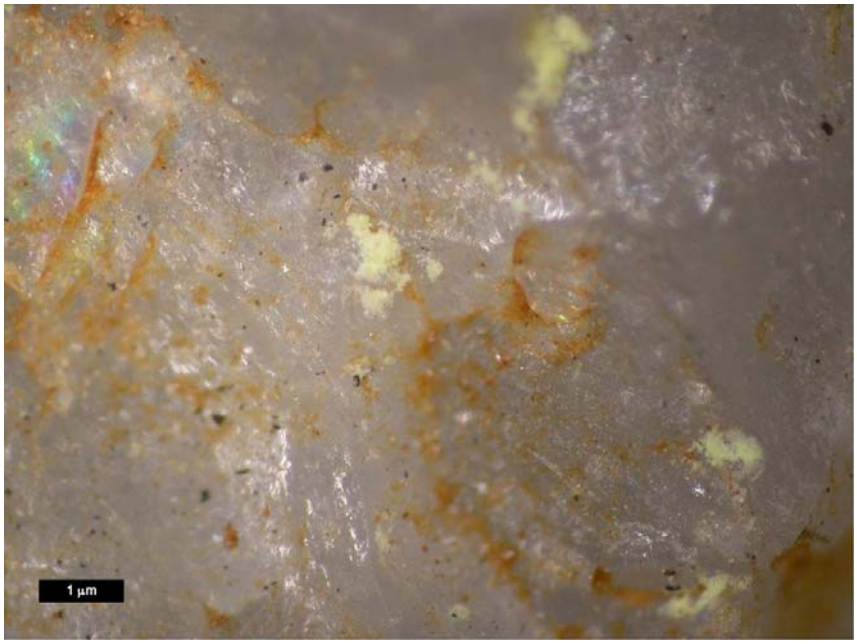


Figure C.54: 7= Micrograph of a green fat-like deposit on STK5872. The contaminant fat-like remains are deposited over sediment. Magnification 100.

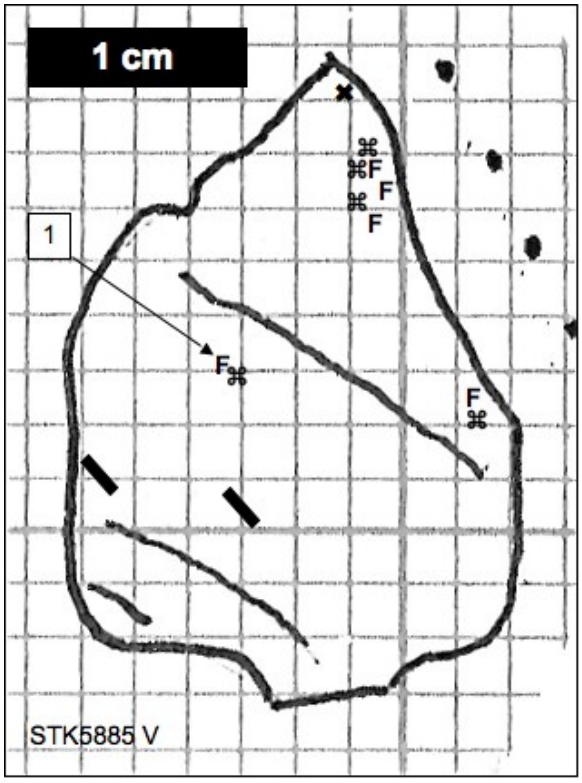


Figure C.55: Distribution map for STK5885, ventral side. Note the broken distal end. For key see Figure C.1.

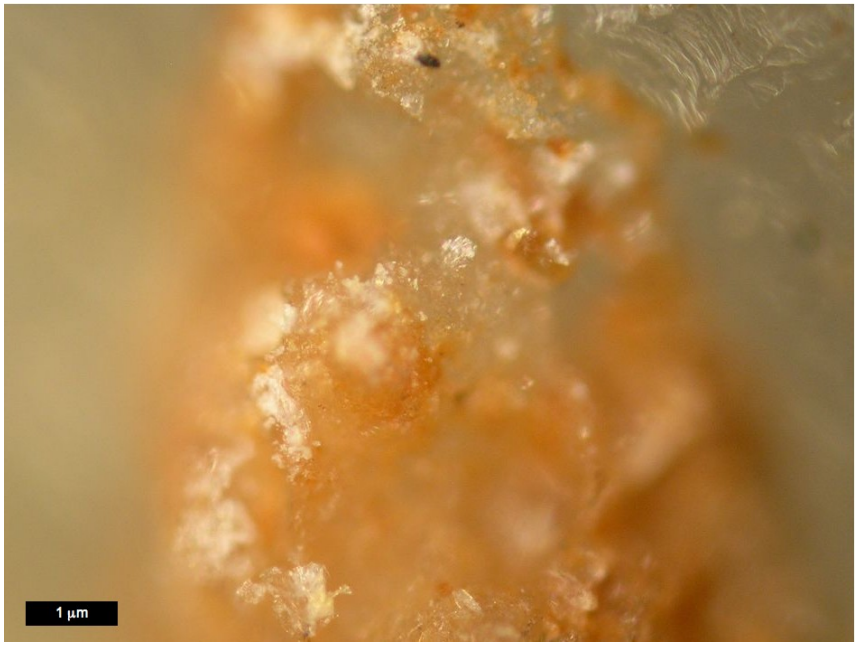


Figure C.56: 1= Micrograph of a fatty bone deposit on STK5885. The fatty bone is deposited on top of orange sediment. Magnification 100.

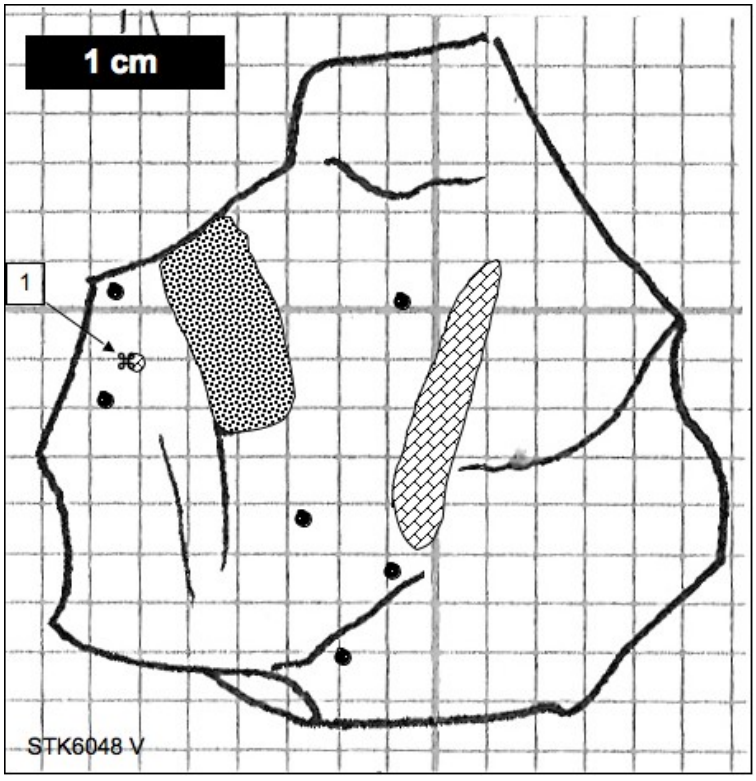


Figure C.57: Distribution map for STK6048, ventral side. For key see Figure C.1.

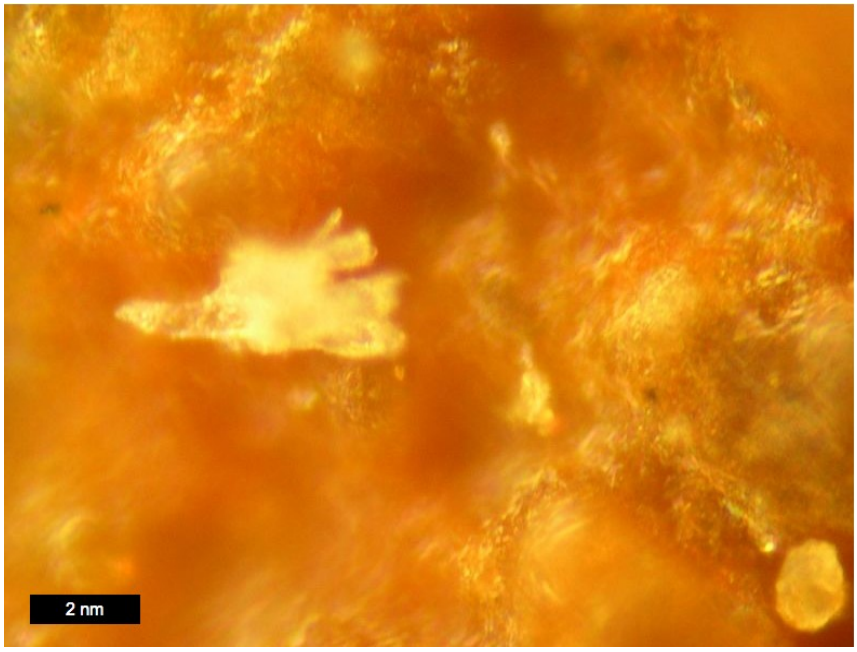


Figure C.58: 1= Micrograph of a connective fibre on STK6048. The fibre is located on top of a sediment patch. Magnification 500.

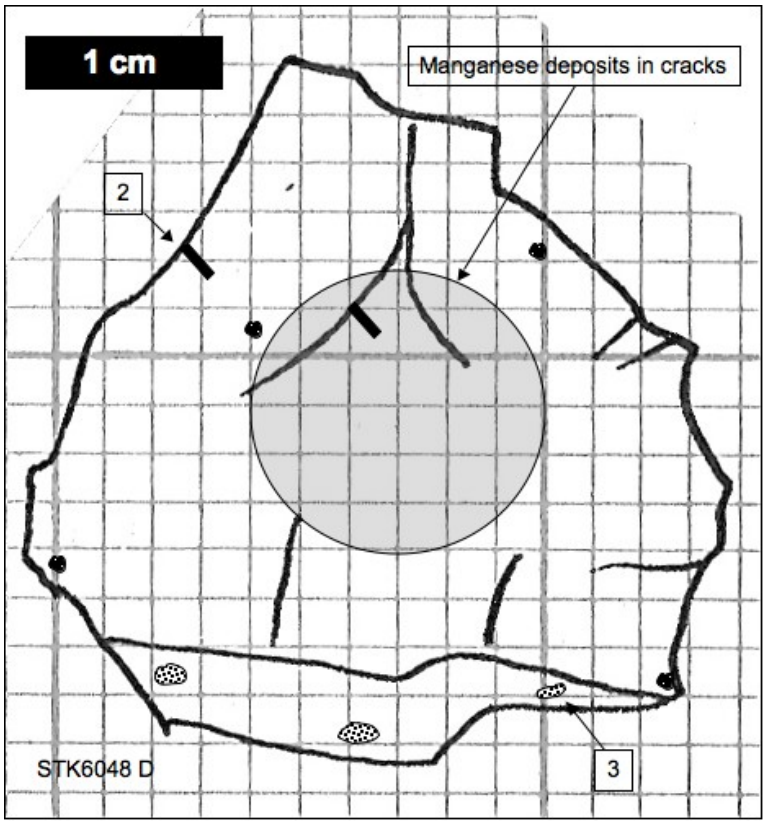


Figure C.59: Distribution map for STK6048, dorsal side. In the cracks inside marked area manganese is abundant. For key see Figure C.1.



Figure C.60: 2= Micrograph of a plant fibre on STK6048. The fibre appears to float over the sediment covered surface. Magnification 50.

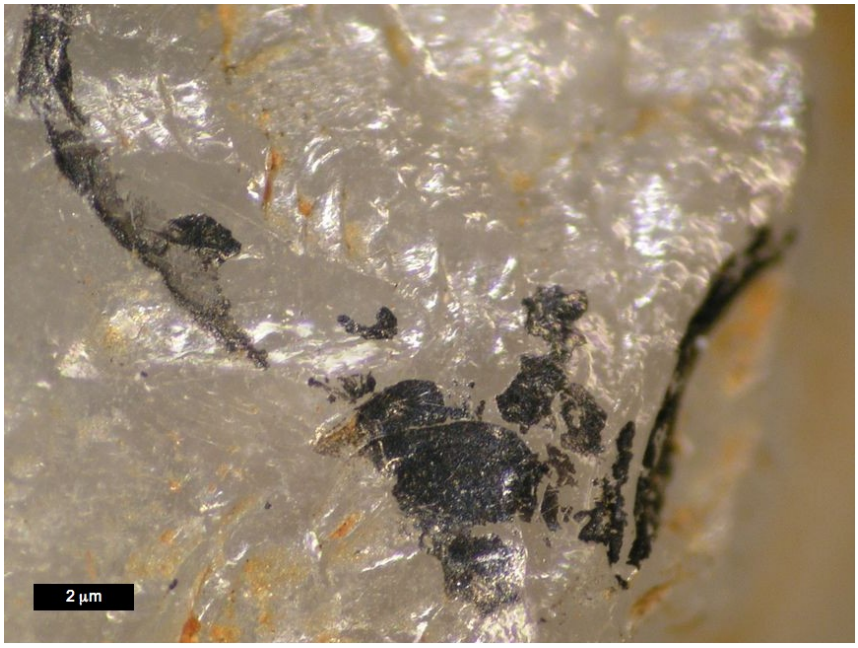


Figure C.61: 3= Micrograph of ink on STK6048. Magnification 50.

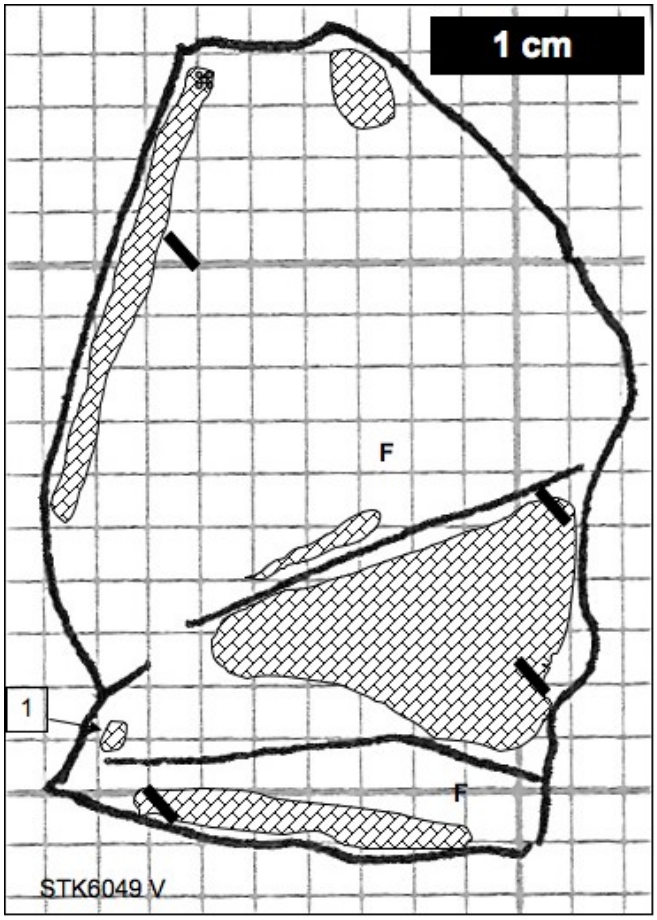


Figure C.62: Distribution map for STK6049, ventral side. For key see Figure C.1.

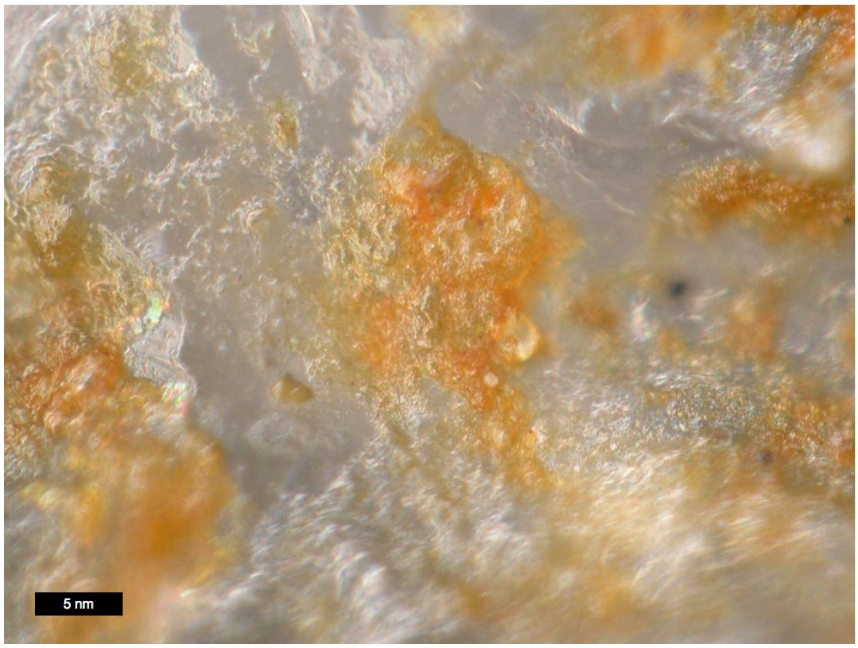


Figure C.63: 1= Micrograph of soil on STK6049. The breccia sediment almost appears liquid. This sediment is present on all the ancient Sterkfontein samples. Magnification 200.

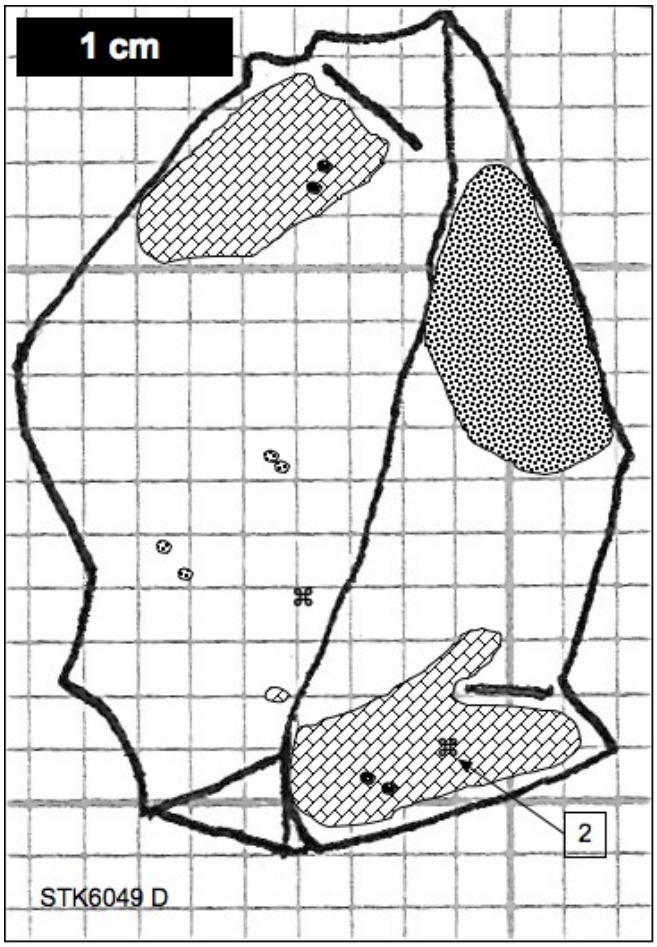


Figure C.64: Distribution map for STK6049, dorsal side. For key see Figure C.1.

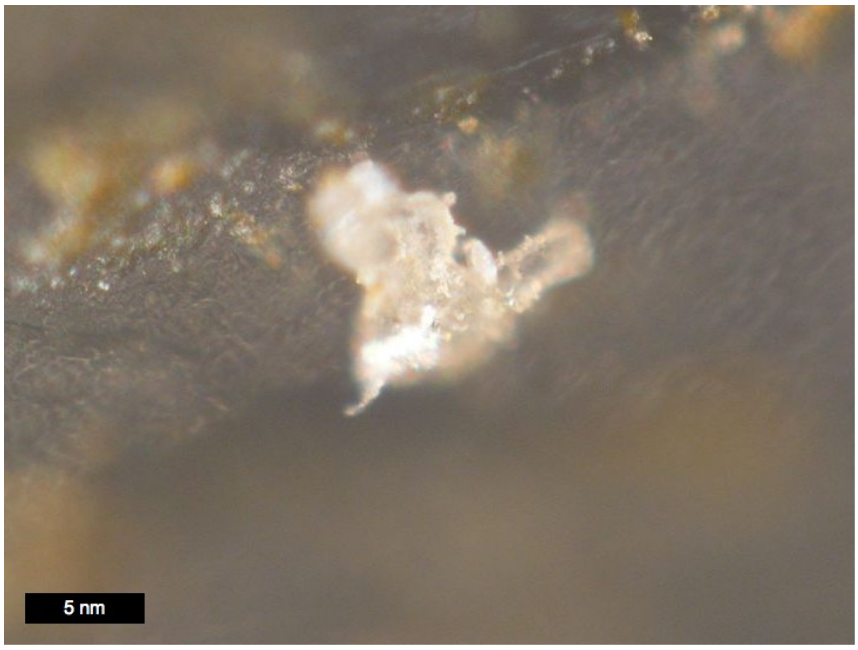


Figure C.65: 2= Micrograph of fatty bone on STK6049. The fatty bone is deposited on top of sediment. Magnification 200.

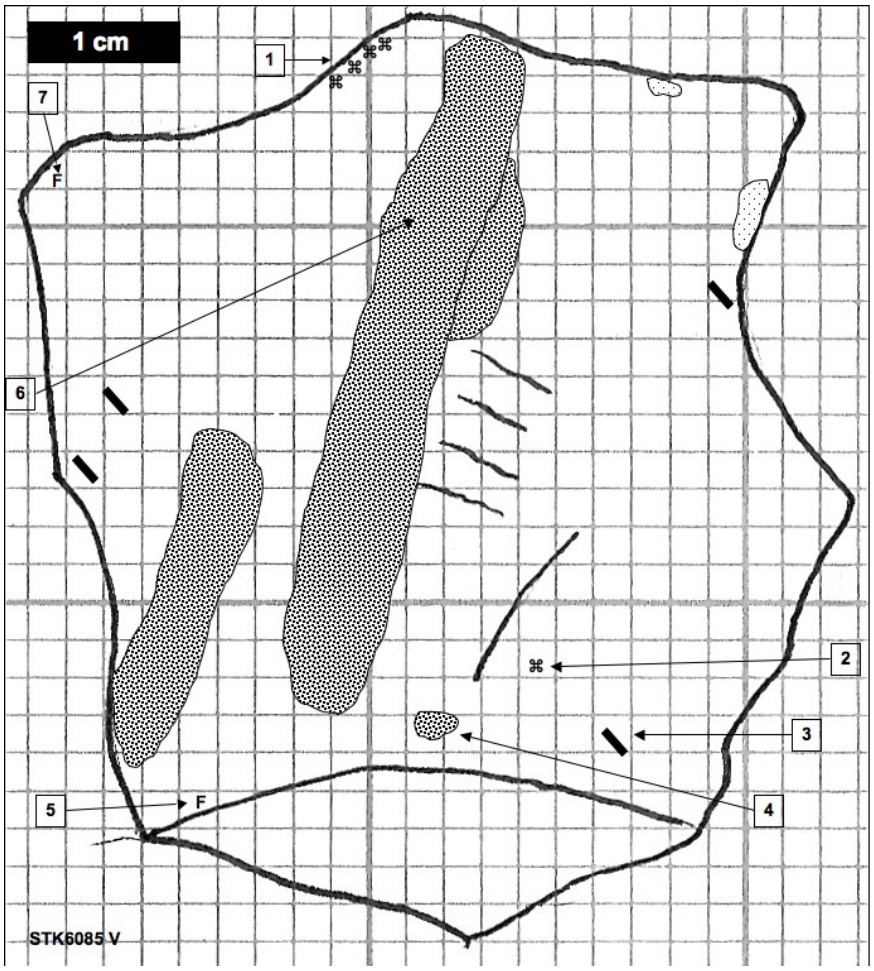


Figure C.66: Distribution map for STK6085, ventral side. Note the large areas with writing. For key see Figure C.1.



Figure C.67: 1= Micrograph of bone on sediment on STK6085. Magnification 200.

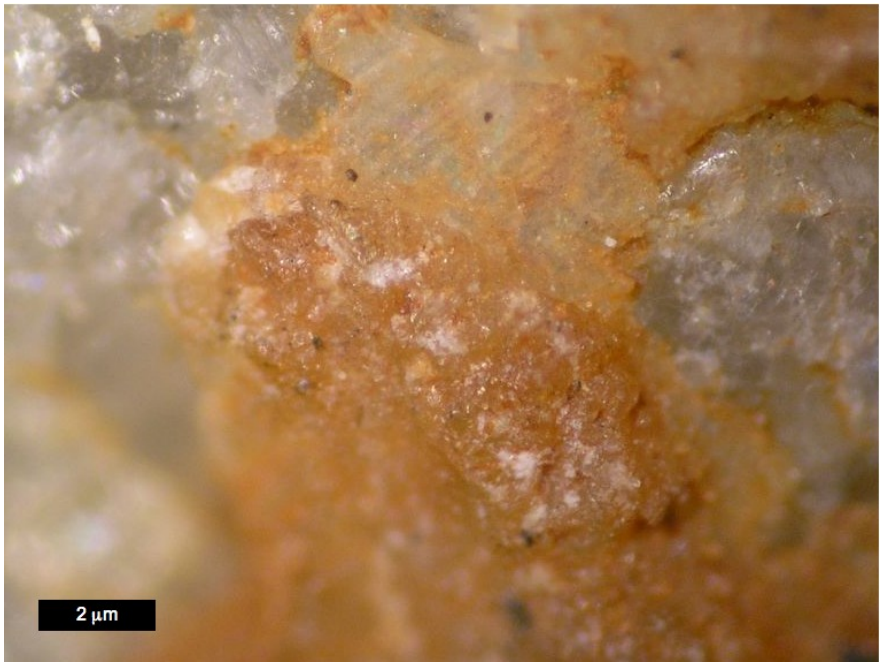


Figure C.68: 2= Micrograph of fatty bone on STK6085. The fatty bone lays on top of a fat deposit. Magnification 50.

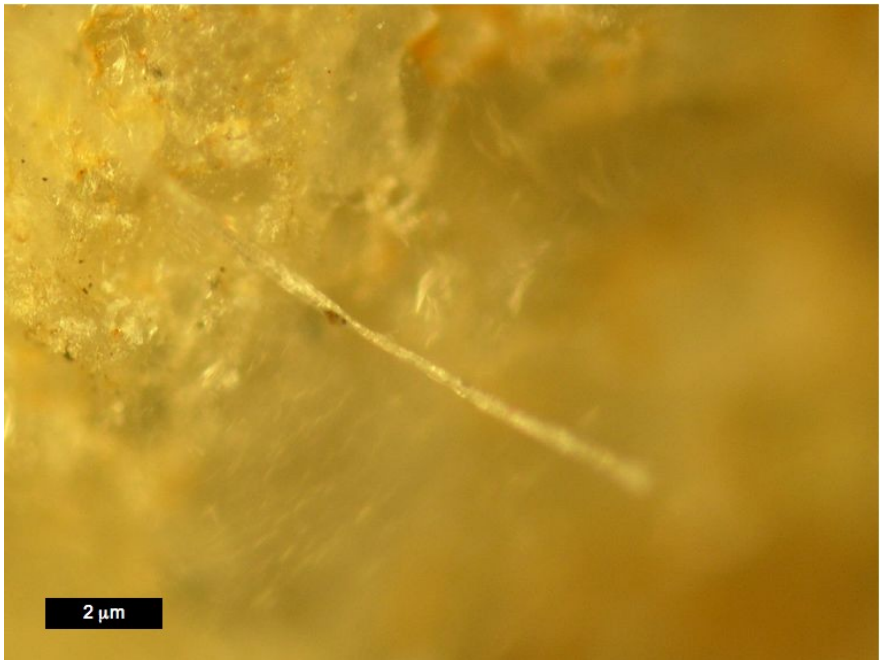


Figure C.69: 3= Micrograph of a fibre on STK6085. Magnification 50.



Figure C.70: 4= Micrograph of ink on STK6085. Magnification 50.



Figure C.71: 5= Micrograph of fat on STK6085. The fat is deposited on top of a sediment patch. Magnification 50.

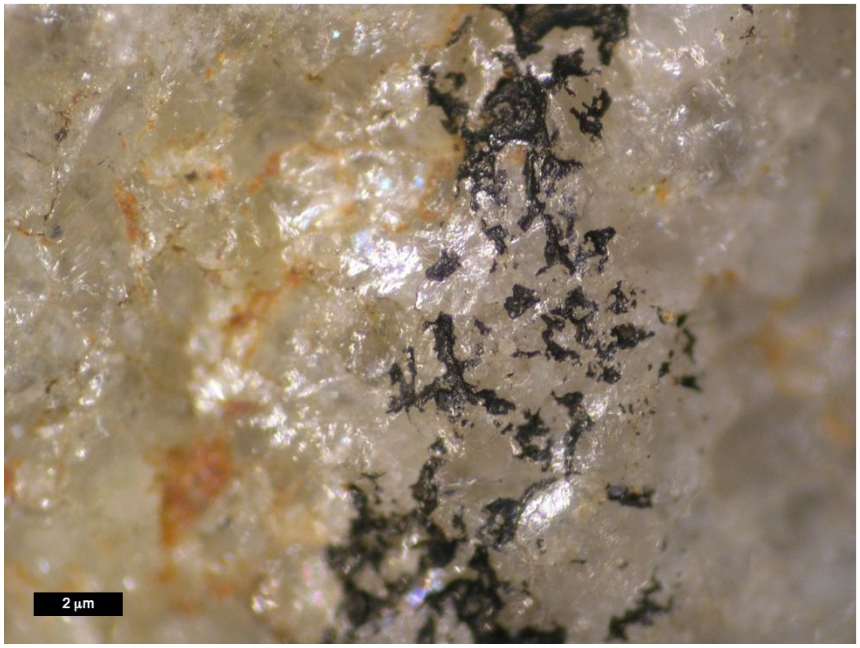


Figure C.72: 6= Micrograph of ink on STK6085. Magnification 50.

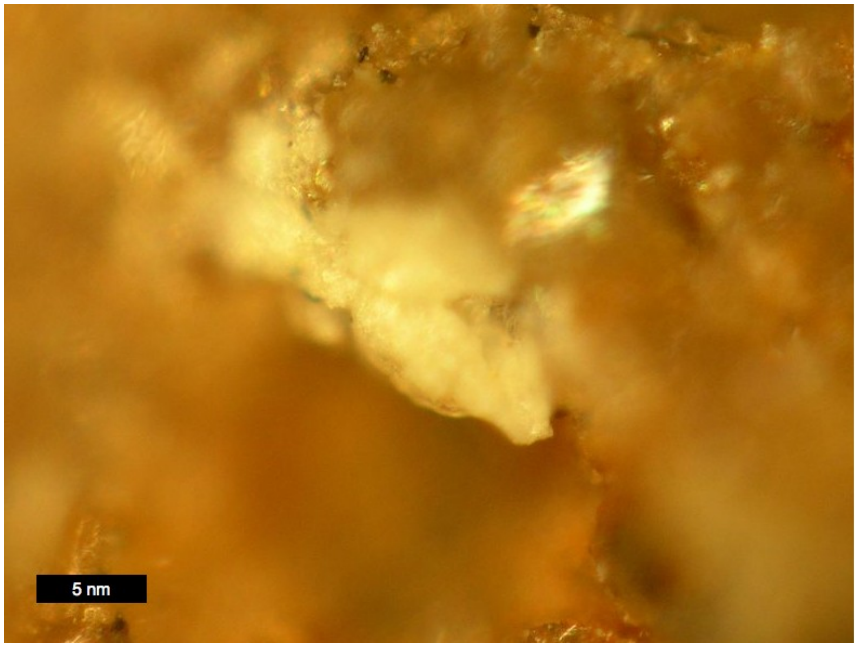


Figure C.73: 7= Micrograph of fat on STK6085. The fat lays on top of a sediment patch. Magnification 200, brightfield polarised light.

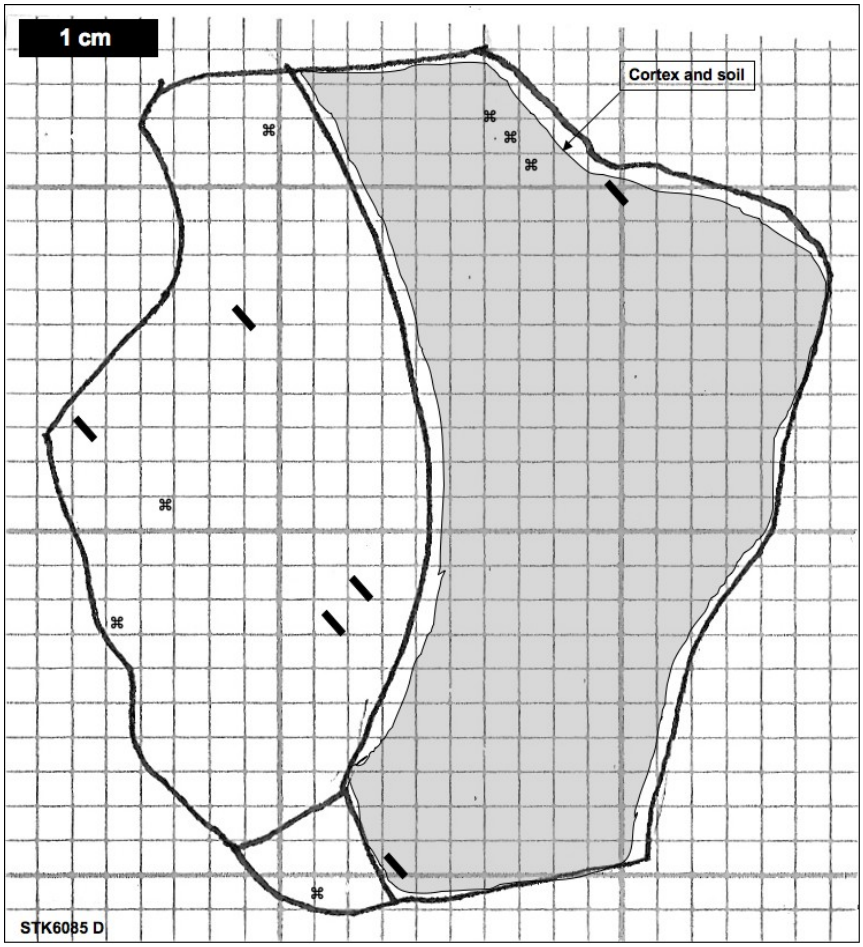


Figure C.74: Distribution map for STK6085, dorsal side. The bone and plant fibres have random distribution. The cortex complicated the identification of soil patches in that area. For key see Figure C.1.

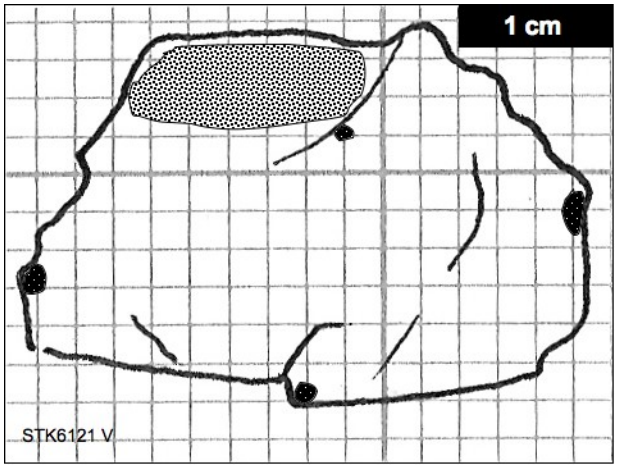


Figure C.75: Distribution map for STK6121, ventral side. For key see Figure C.1.

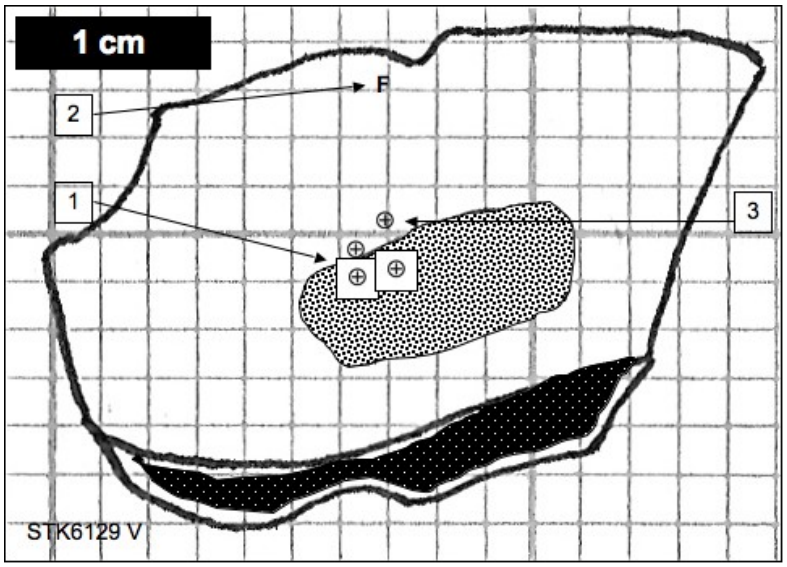


Figure C.76: Distribution map for STK6129, ventral side. For key see Figure C.1.

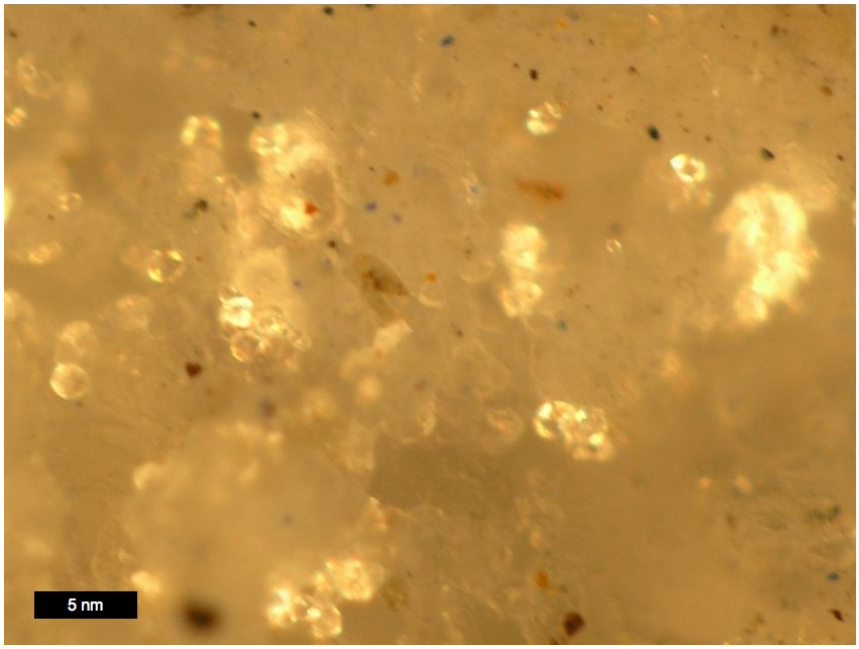


Figure C.77: 1= Micrograph of starch grains on STK6129. The starch grains lay on top of the nail varnish label. Magnification 200, brightfield polarised light.

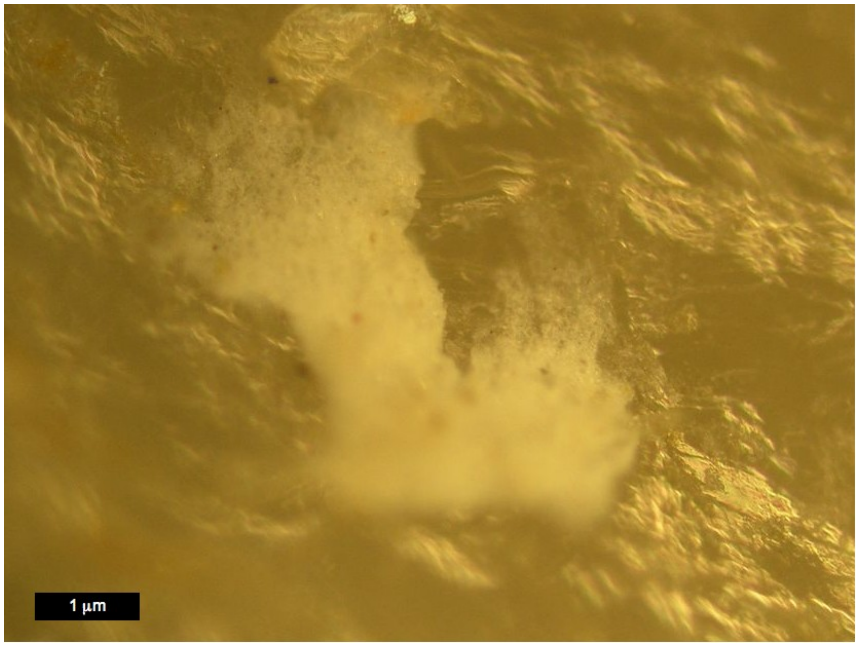


Figure C.78: 2= Micrograph of fat on STK6129. This smeared deposit is the only animal type residue on this artefact. Magnification 100.

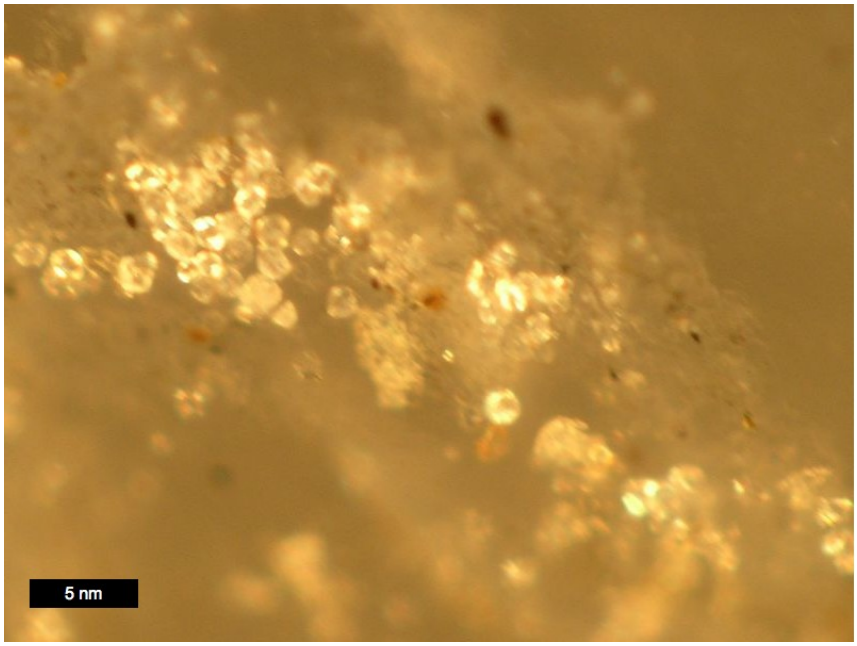


Figure C.79: 3= Micrograph of starch grains on STK6129. Magnification 200, brightfield polarised light.

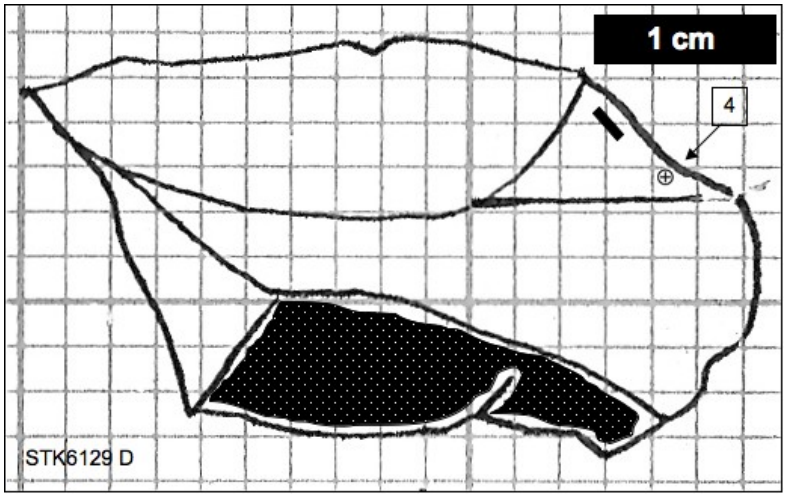


Figure C.80: Distribution map for STK6129, dorsal side. For key see Figure C.1.

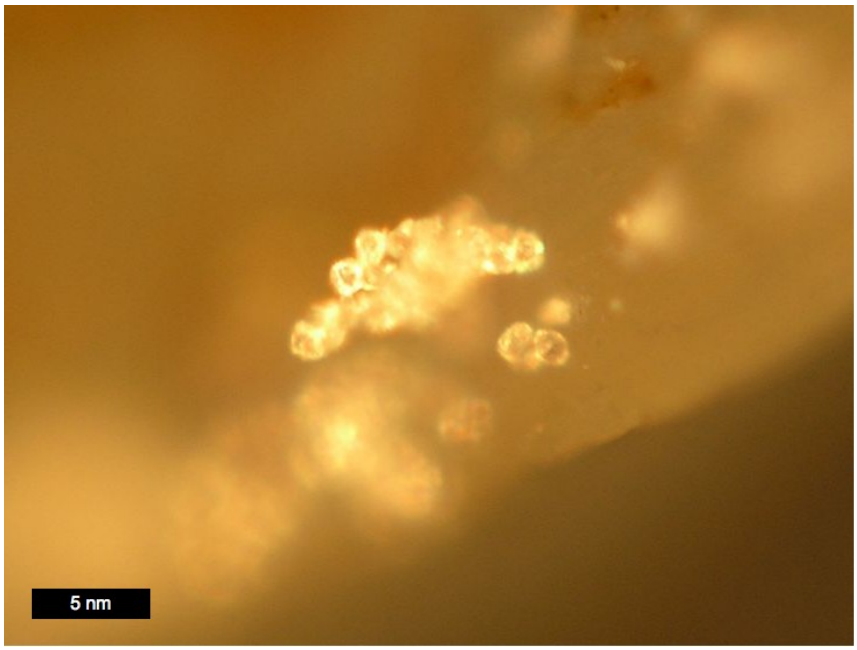


Figure C.81: 4= Micrograph of starch grains on STK6129. Because starch grains are also located on top of the nail varnish label, these starch grains are disregarded. Magnification 200, brightfield polarised light.

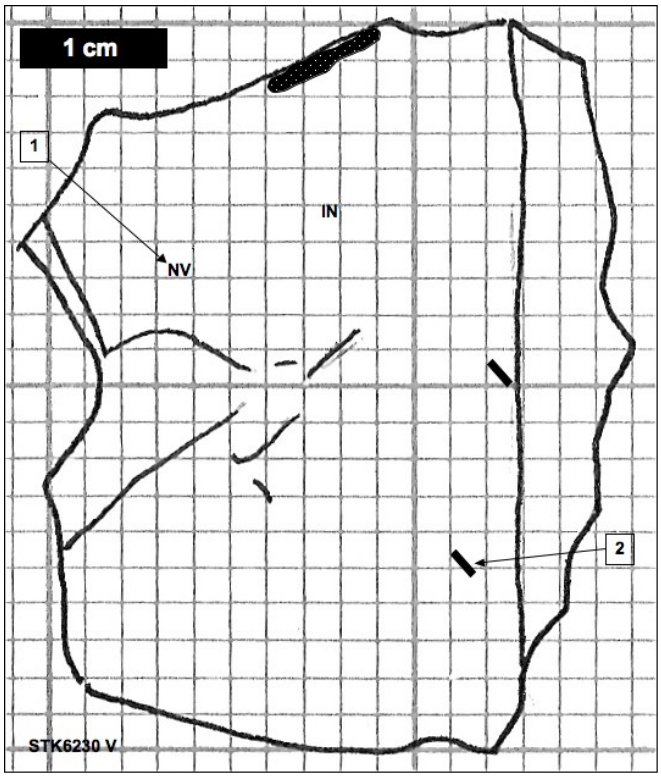


Figure C.82: Distribution map for STK6230, ventral side. For key see Figure C.1.

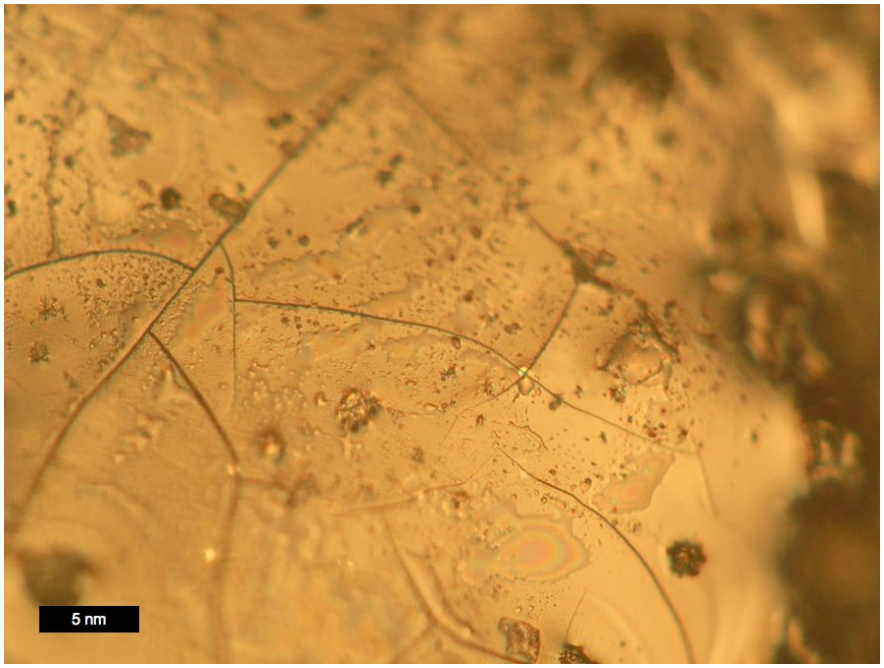


Figure C.83: 1= Micrograph of nail varnish on STK6230. Magnification 200.

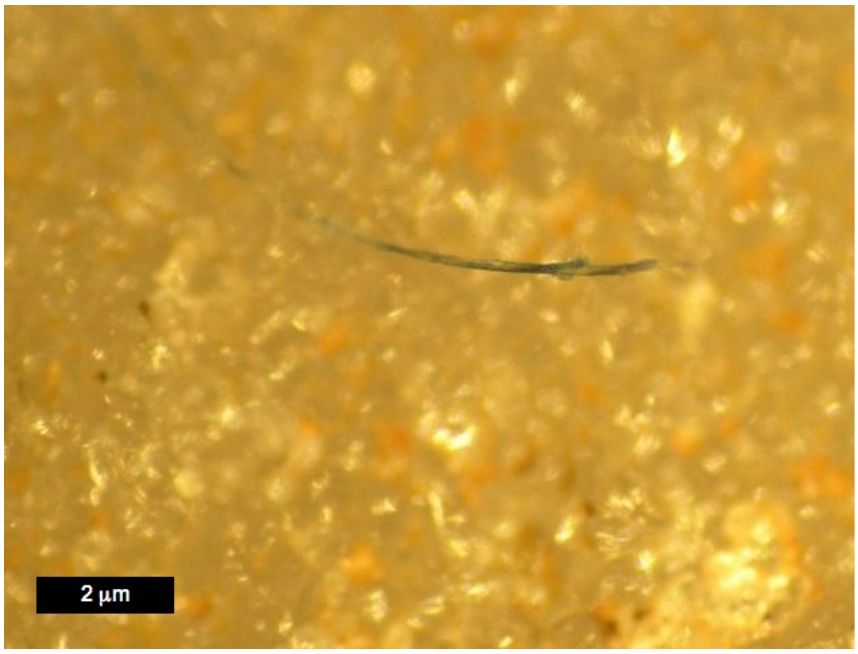


Figure C.84: 2= Micrograph of a blue fibre on STK6230. This blue fibre is similar to the modern fibre in Figure A.11. Magnification 50.

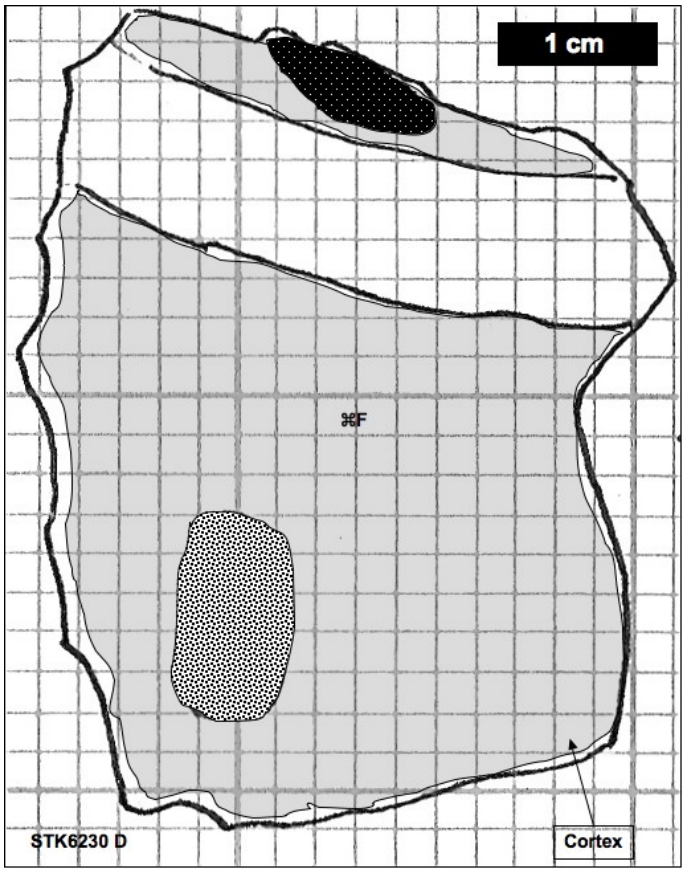


Figure C.85: Distribution map for STK6230, dorsal side. For key see Figure C.1.

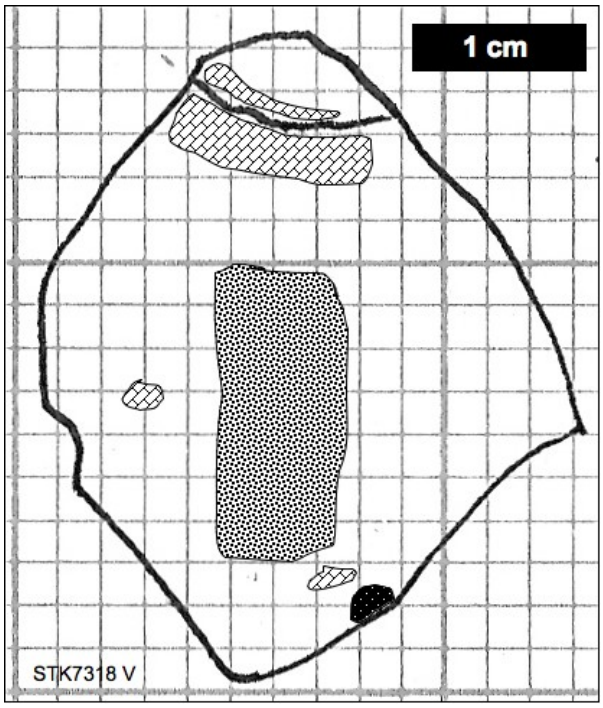


Figure C.86: Distribution map for STK7318, ventral side. For key see Figure C.1.

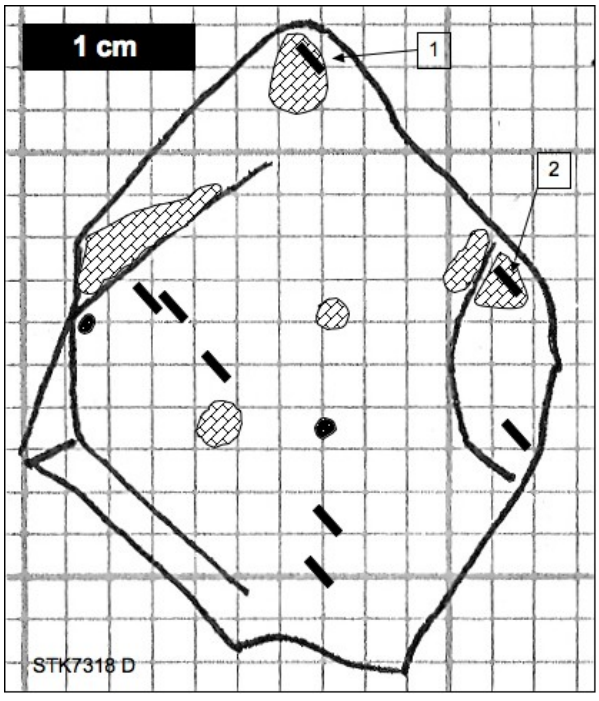


Figure C.87: Distribution map for STK7318, dorsal side. For key see Figure C.1.

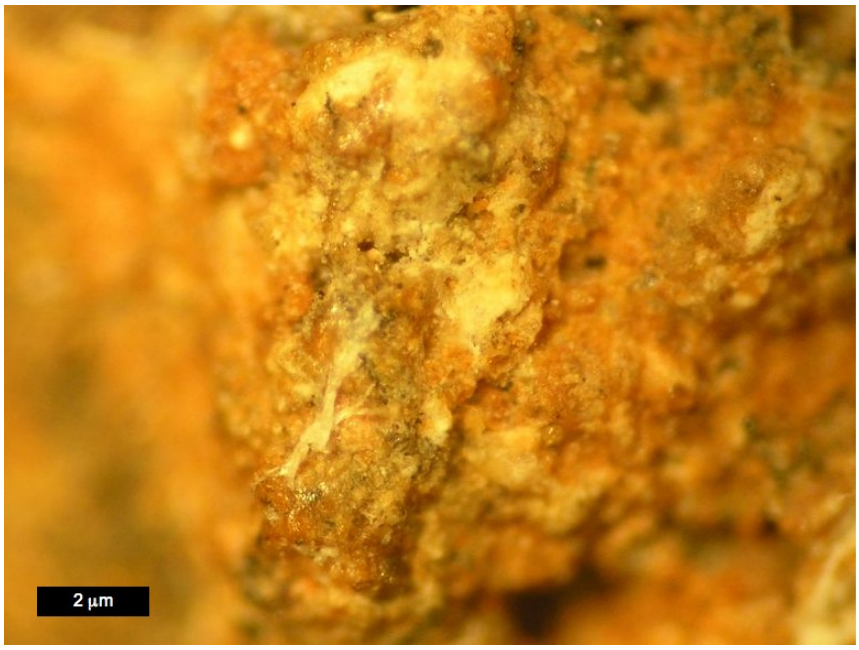


Figure C.88: 1= Micrograph of a plant fibre on STK7318. The fibre lays on top of a sediment patch. Magnification 50.



Figure C.89: 2= Micrograph of a plant fibre on STK7318. The fibre is located on top of a sediment patch. Because this fibre and the one in C.88 are clear contaminants, the remaining plant fibres are compromised. Magnification 50.

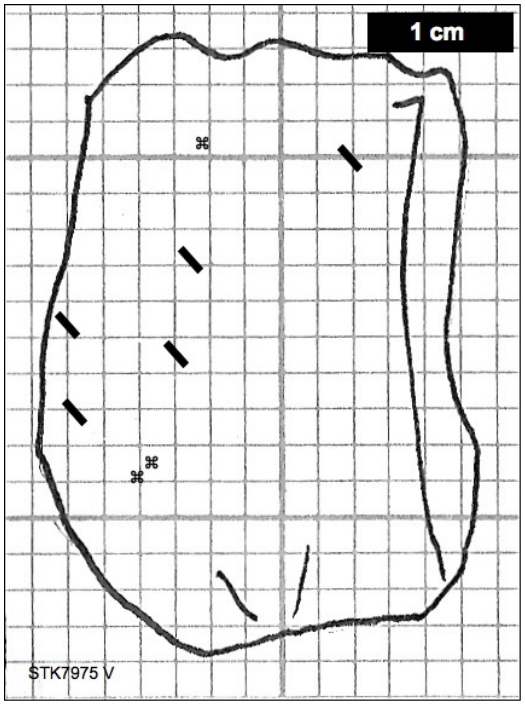


Figure C.90: Distribution map for STK7975, ventral side. For key see Figure C.1.

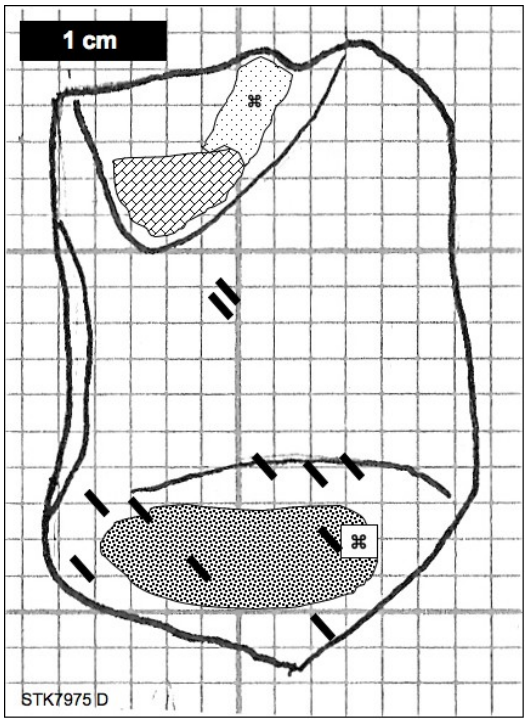


Figure C.91: Distribution map for STK7975, dorsal side. The plant fibres and bone deposit are located on top of ash and the nail varnish label and therefore disregarded. In addition, the plant fibres have an arbitrary distribution. For key see Figure C.1.

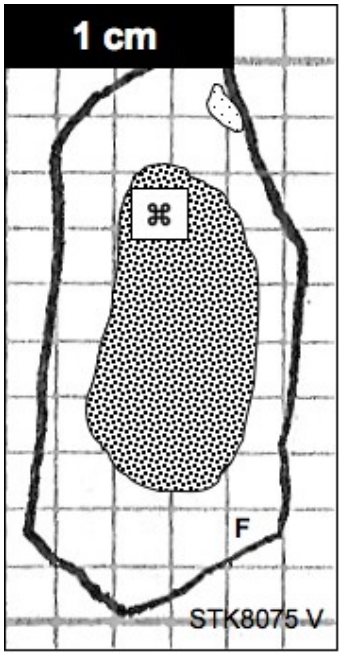


Figure C.92: Distribution map for STK8075, ventral side. For key see Figure C.1.



Figure C.93: Photo of STK8849, ventral side. Note the ink and writing.

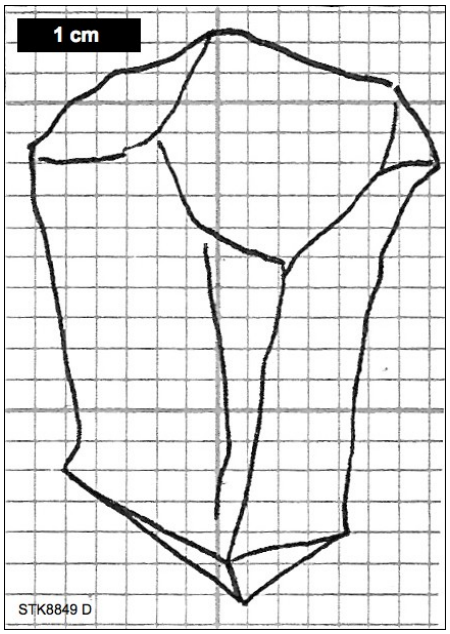


Figure C.94: Distribution map for STK8849, dorsal side. For key see Figure C.1.

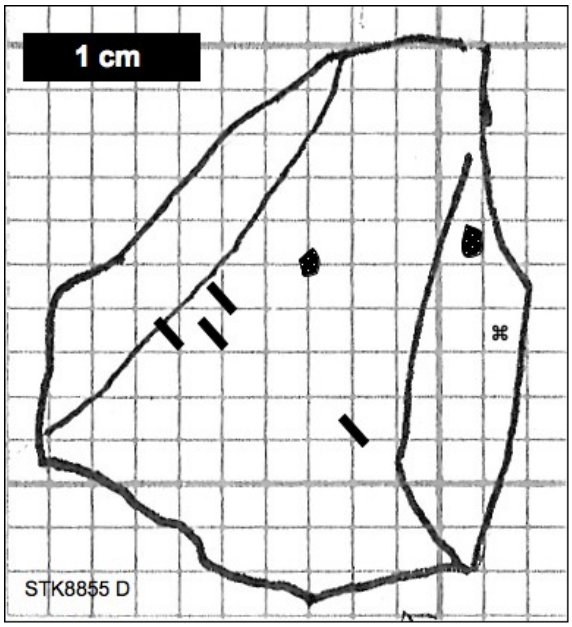


Figure C.95: Distribution map for STK8855, dorsal side. For key see Figure C.1.

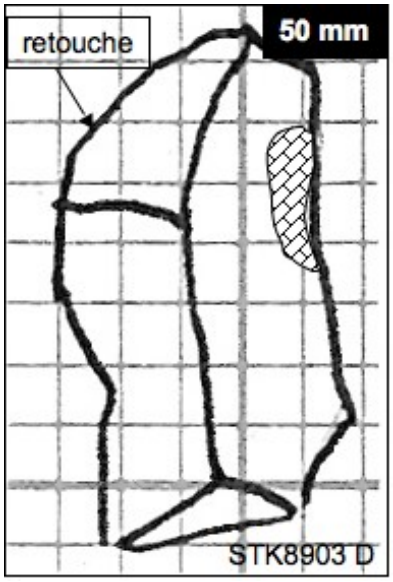


Figure C.96: Distribution map for STK8903, dorsal side. For key see Figure C.1.

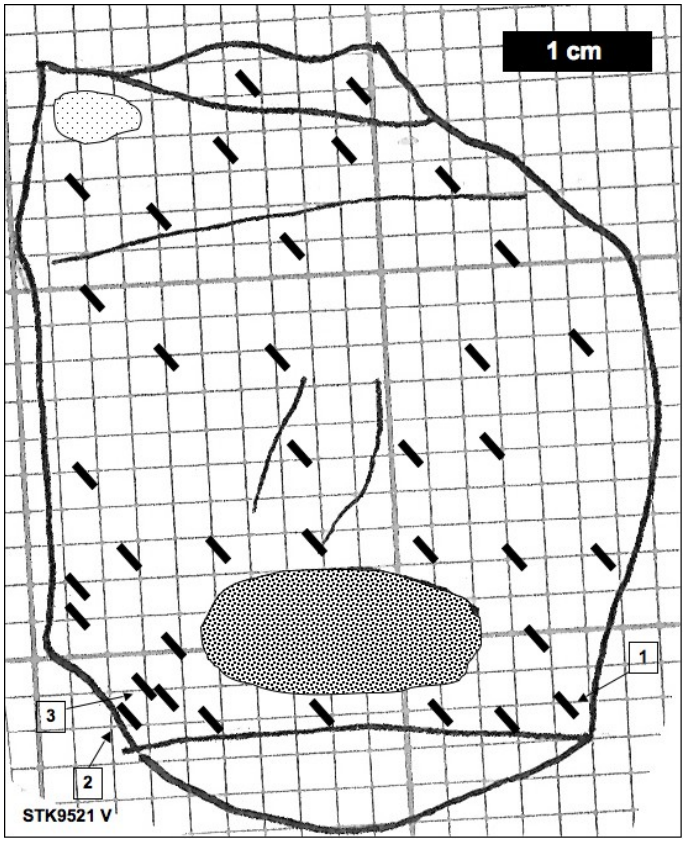


Figure C.97: Distribution map for STK9521, ventral side. Note the uniform distribution of plant fibres. This is indicative for contaminants. For key see Figure C.1.



Figure C.98: 1= Micrograph of plant fibres on STK9521. The fibres lay o top sediment patches. All the fibres on this artefact are extremely similar. Magnification 100.



Figure C.99: 2= Micrograph of plant fibres on STK9521. The fibres lay on top of sediment. Magnification 100.



Figure C.100: 3= Micrograph of plant fibres on STK9521. The fibres lay on top sediment. Magnification 50.

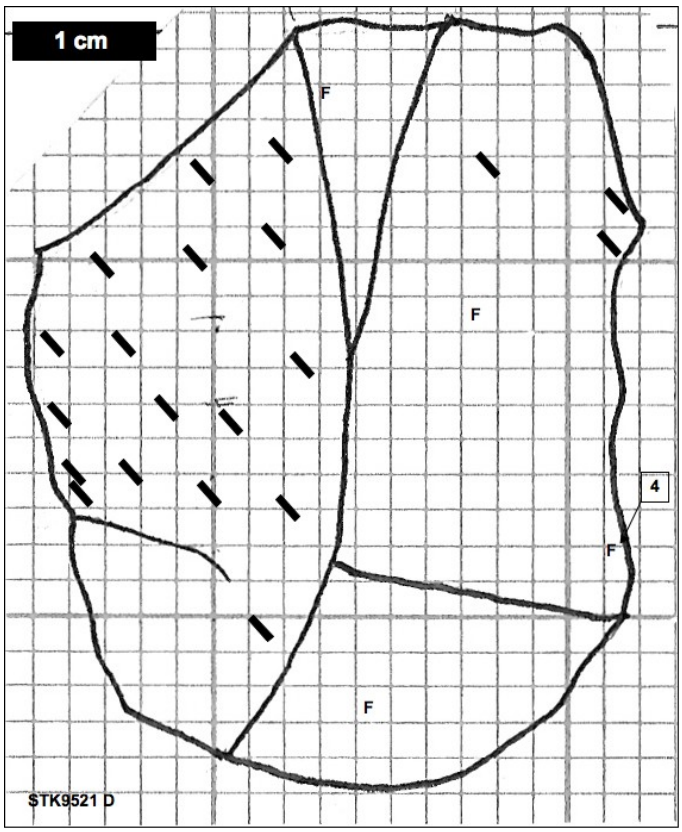


Figure C.101: Distribution map for STK9521, dorsal side. For key see Figure C.1.

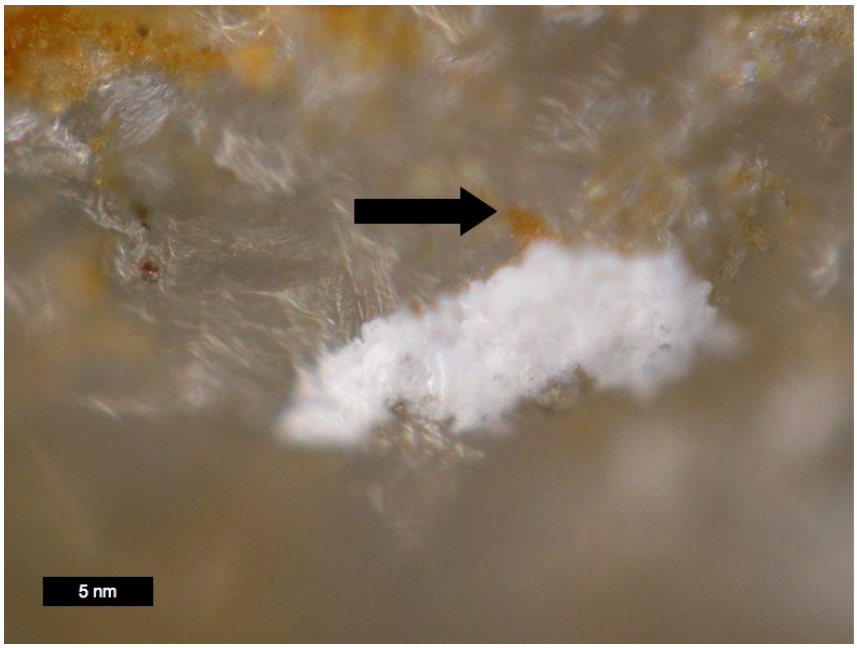


Figure C.102: 4= Micrograph of fat on STK9521. Note that the fat-like deposit overlies a sediment patch (indicated by the arrow). Magnification 200.

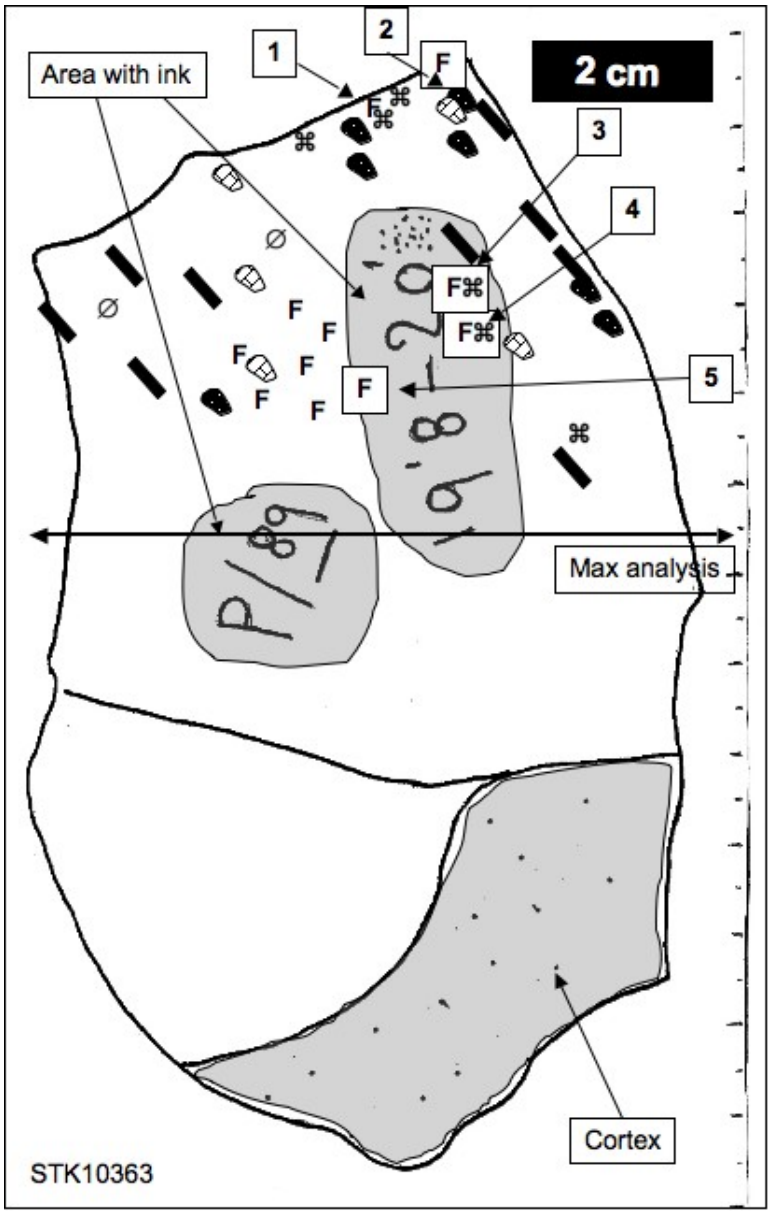


Figure C.103: Distribution map for STK10363. For key see Figure C.1.

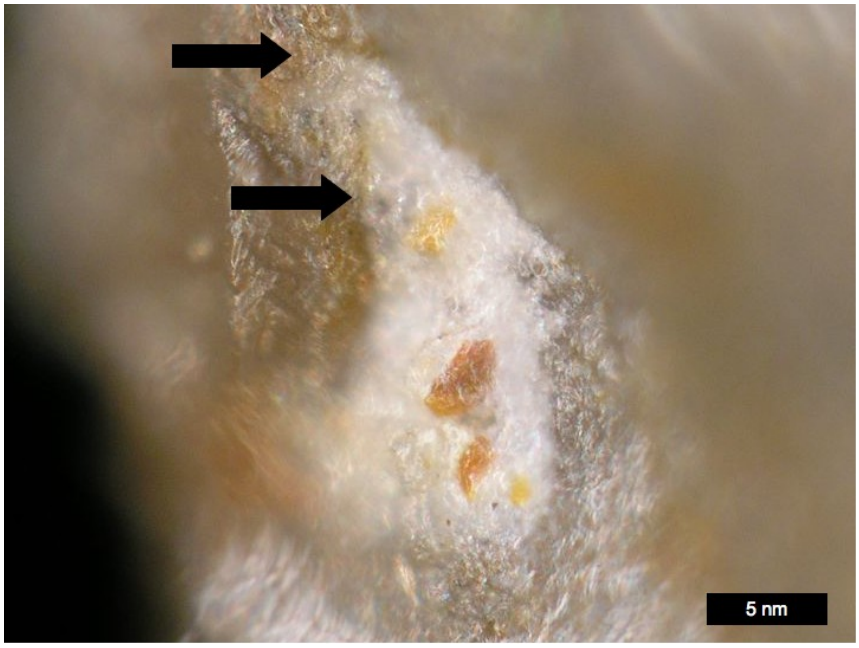


Figure C.104: 1= Micrograph of fat on STK10363. Note that the fat overlies a thin sediment layer (indicated with the arrows). Magnification 200.

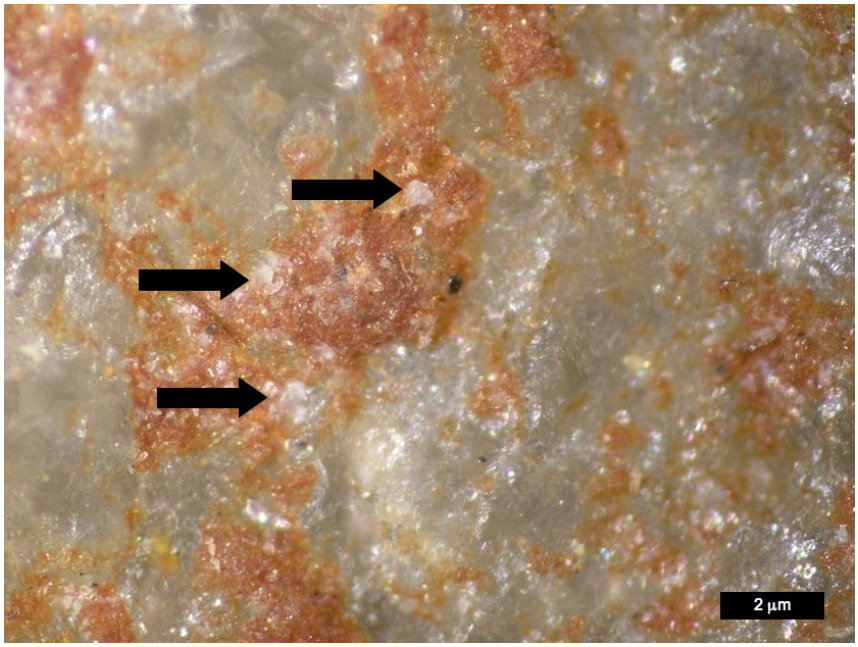


Figure C.105: 2= Micrograph of sediment with fat-like deposits on top (indicated with the arrows) on STK10363. Magnification 50.

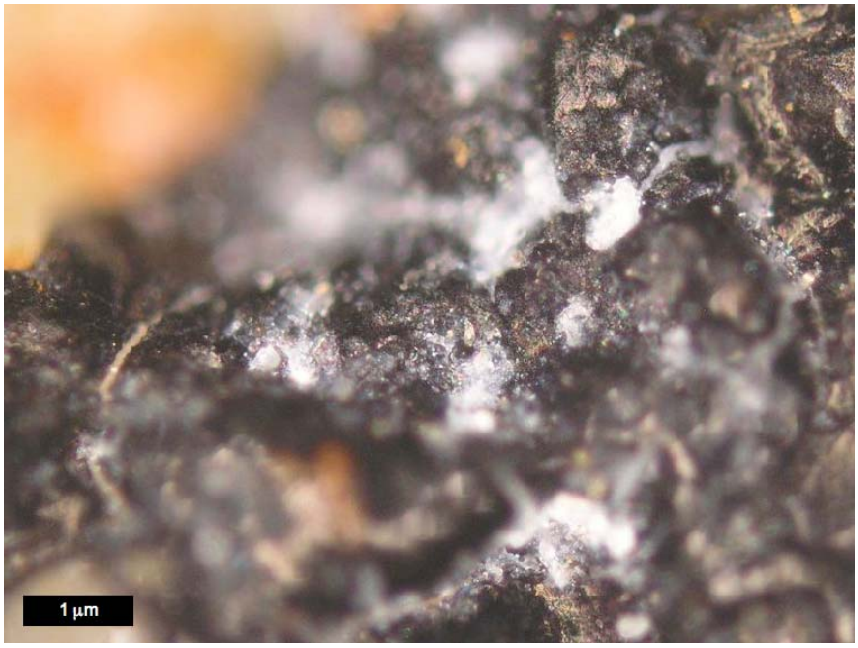


Figure C.106: 3= Micrograph of fatty bone on STK10363. Note that the fatty bone is deposited on top of the label. Magnification 100.

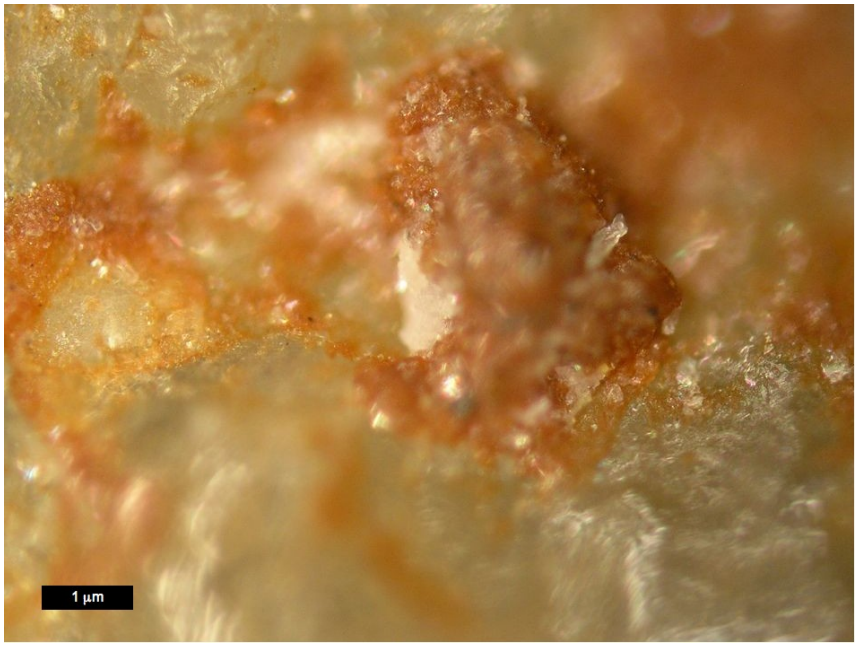


Figure C.107: 4= Micrograph of fatty bone on STK10363. The white fatty bone-like residues lay on top of an orange sediment patch. Magnification 100.

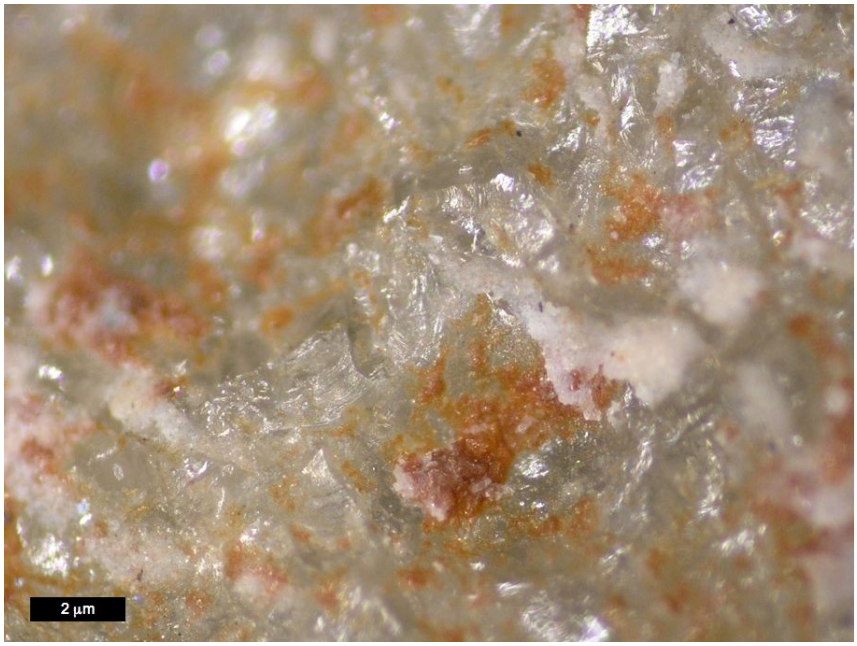


Figure C.108: 5= Micrograph of a fat-like deposit on STK10363. The fat is deposited over a sediment patch. Magnification 50.

D APPENDIX: RESULTS FROM THE ANCIENT SIBUDU ARCHAEOLOGICAL RESULTS

This appendix presents the results from Chapter Six: the Sibudu archaeological results. The appendix consists of micrographs and distribution maps on which the locations of all residues are indicated per stone tool. Unless stated otherwise all micrographs are taken in darkfield light using no filters. The key for the distribution maps is as follows:

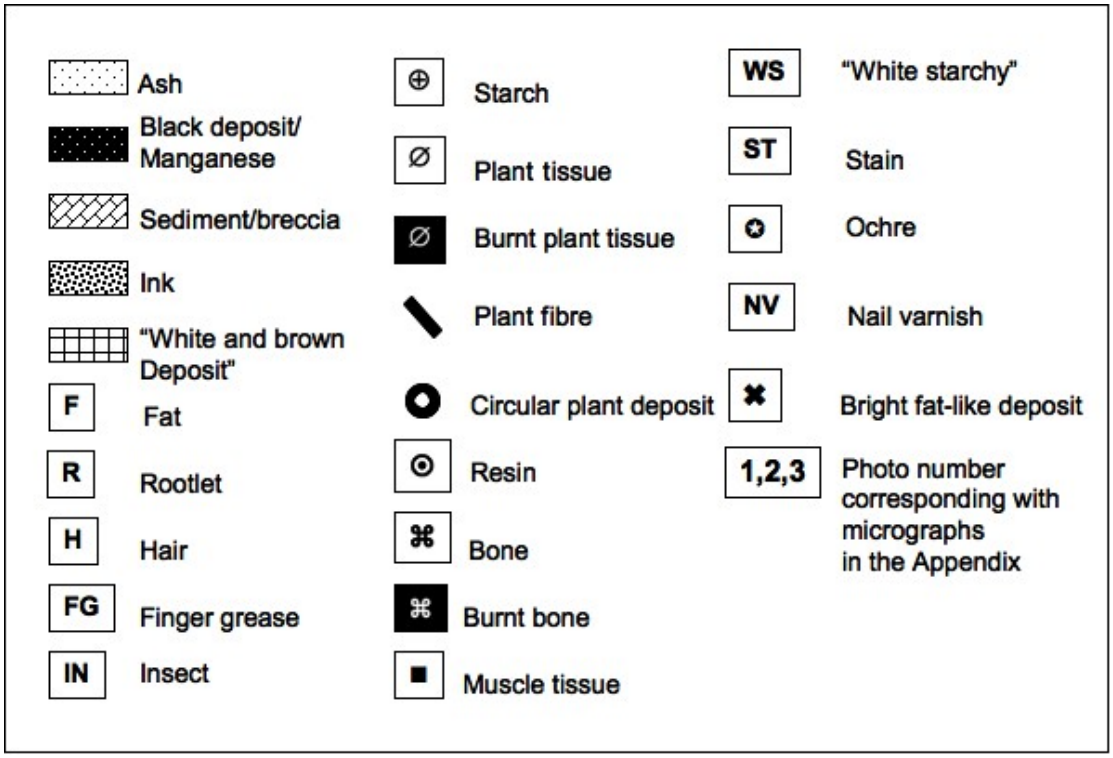


Figure D.1: Key to figures.

Some maps have arrows and numbers, which indicate the location of micrographs. These micrographs are presented underneath the distribution map and start with the assigned map number. All the tools in the Figures are orientated with the distal end facing upwards.

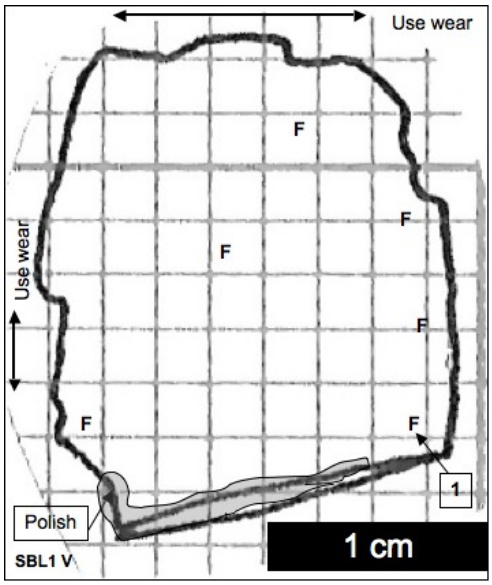


Figure D.2: Distribution map for SBL1, ventral side. For key see Figure D.1.

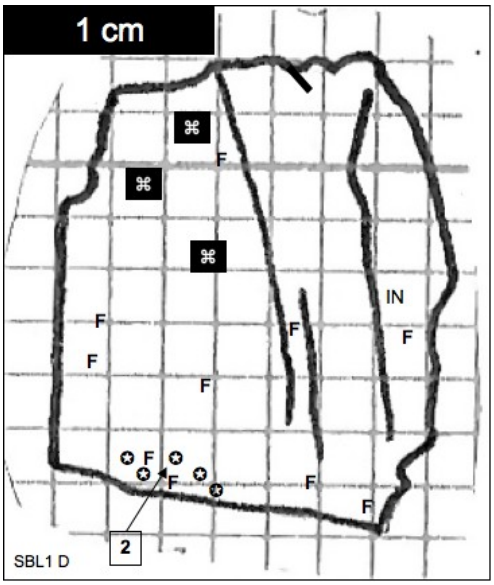


Figure D.3: Distribution map for SBL1, dorsal side. For key see Figure D.1.



Figure D.4: 1= Micrograph of fat on SBL1. Magnification 200.

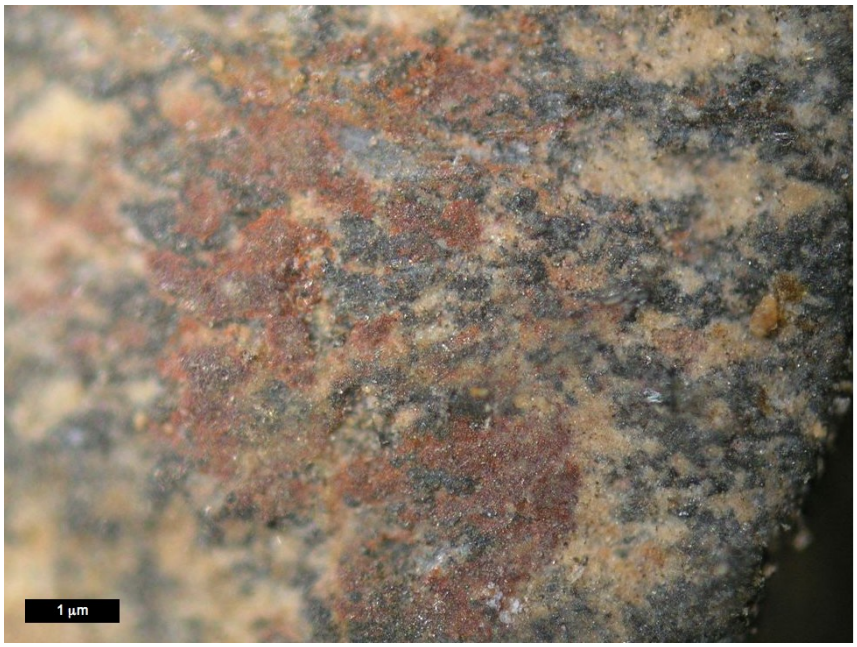


Figure D.5: 2= Micrograph of red ochre and white/yellow fat on SBL1. Magnification 100.

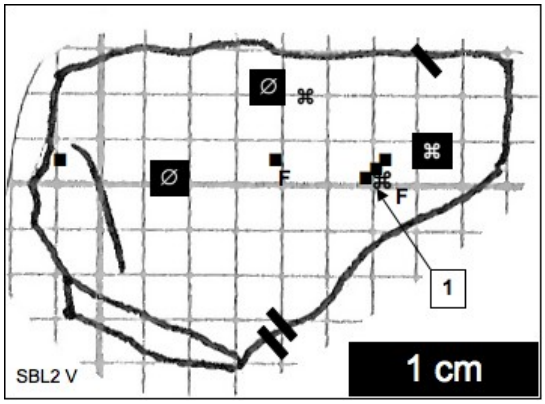


Figure D.6: Distribution map for SBL2, ventral side. For key see Figure D.1. Note that there is a suite of animal type residues present on this artefact.

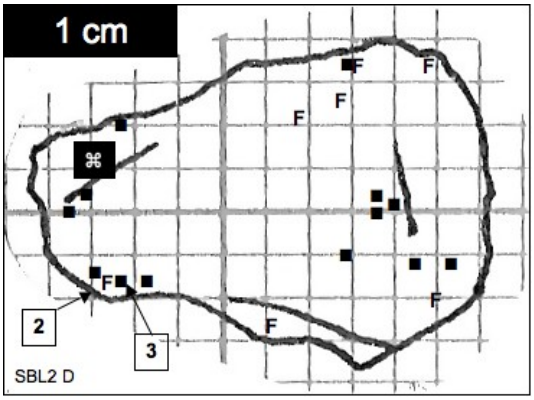


Figure D.7: Distribution map for SBL2, dorsal side. For key see Figure D.1. Note that there is a suite of animal type residues present on this artefact.

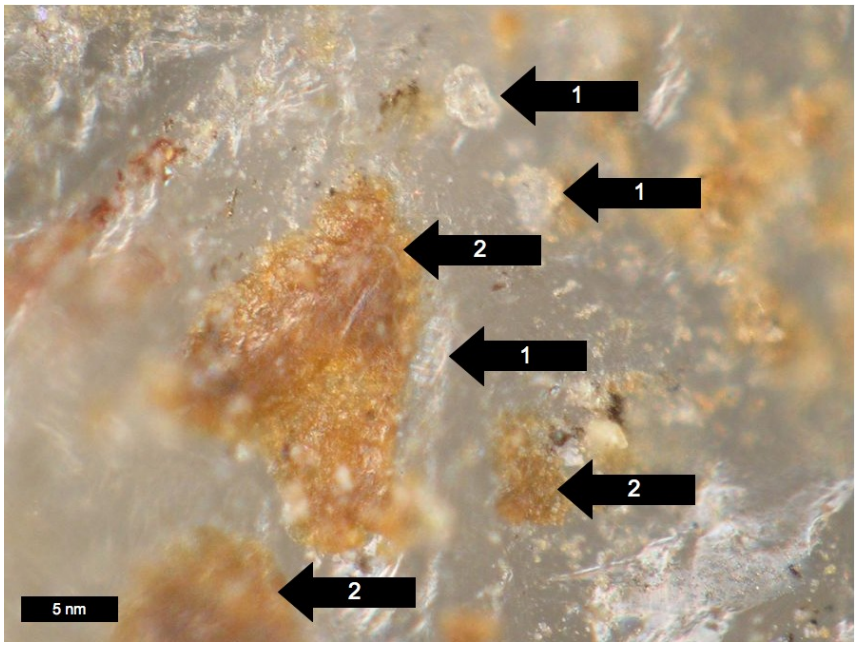


Figure D.8: 1= Micrograph of bone (arrow 1) and muscle tissue (arrow 2) on SBL2. Magnification 200.



Figure D.9: 2= Micrograph of white fat on soil on SBL2. Magnification 100.

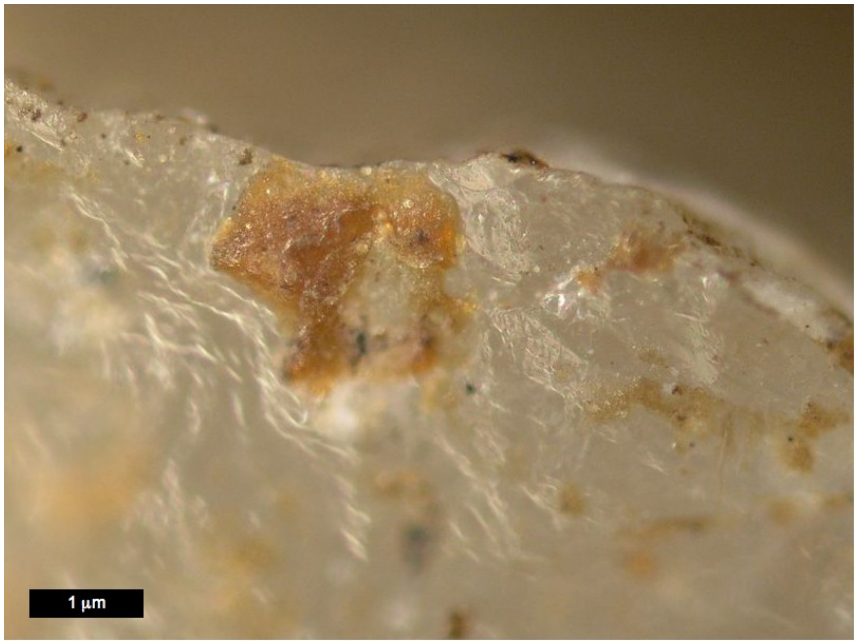


Figure D.10: 3= Micrograph of muscle tissue on SBL2. Magnification 100.

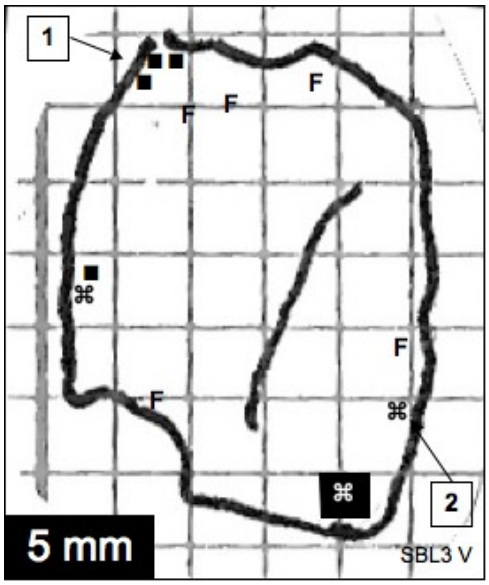


Figure D.11: Distribution map for SBL3, ventral side. Note that there is a suite of animal type residues present on this artefact. For key see Figure D.1.

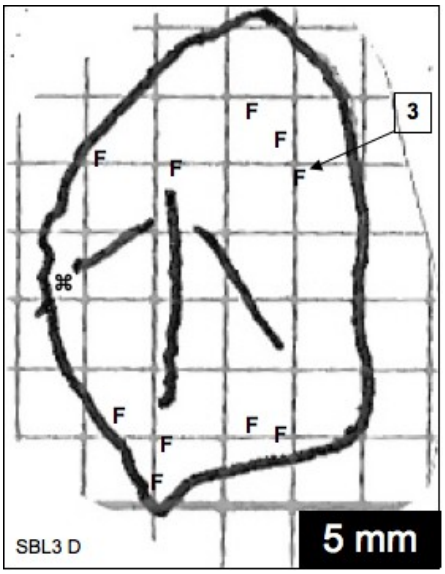


Figure D.12: Distribution map for SBL3, dorsal side. For key see Figure D.1.

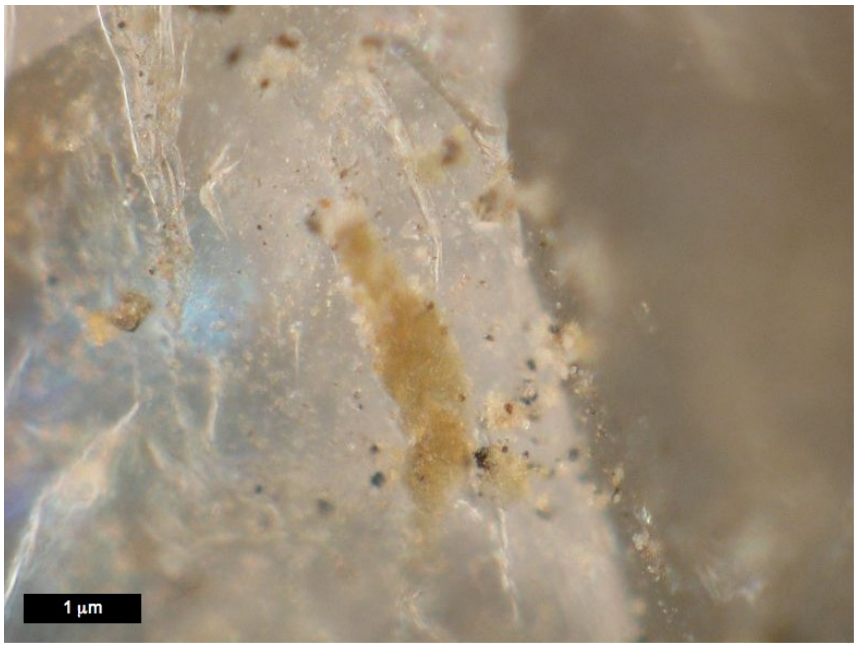


Figure D.13: 1= Micrograph of a thin muscle tissue deposit on SBL3. Magnification 100.

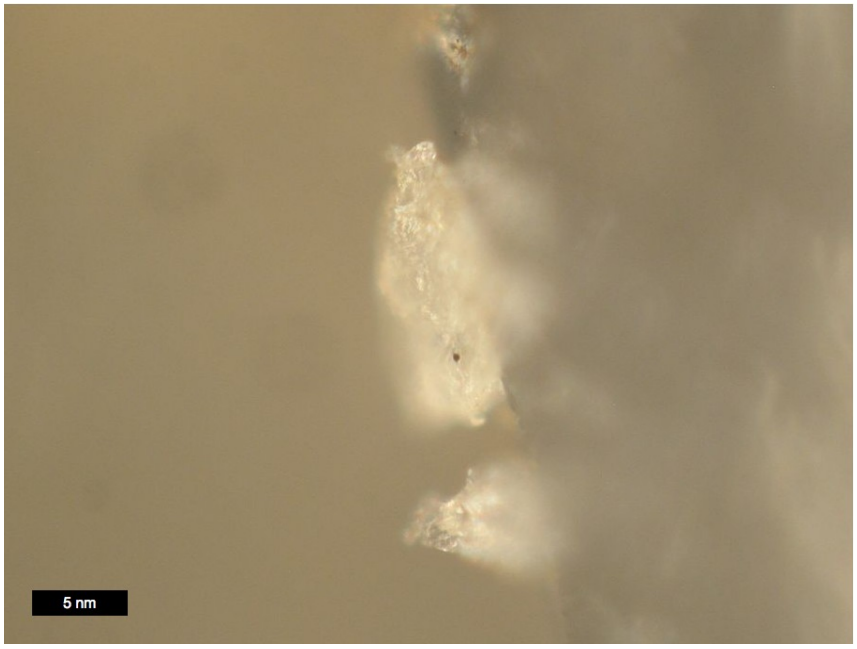


Figure D.14: 2= Micrograph of fatty bone on SBL3. Magnification 200.

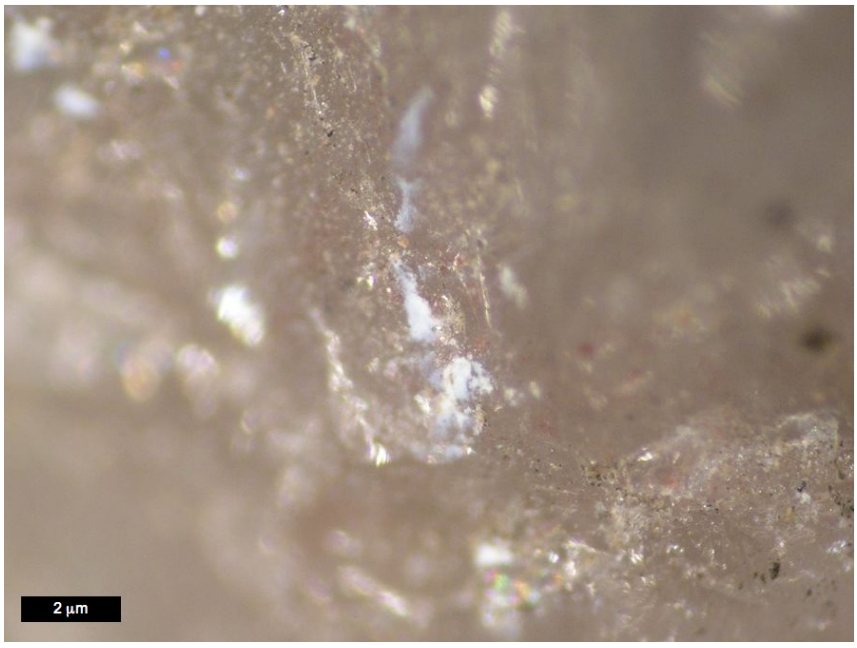


Figure D.15: 3= Micrograph of bluish fat on SBL3. Magnification 50.

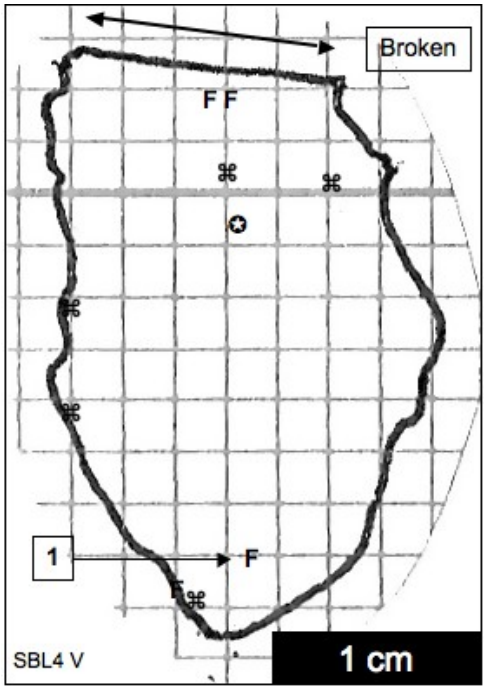


Figure D.16: Distribution map for SBL4, ventral side. Note that the bone deposits are mainly located around the edge. For key see Figure D.1.

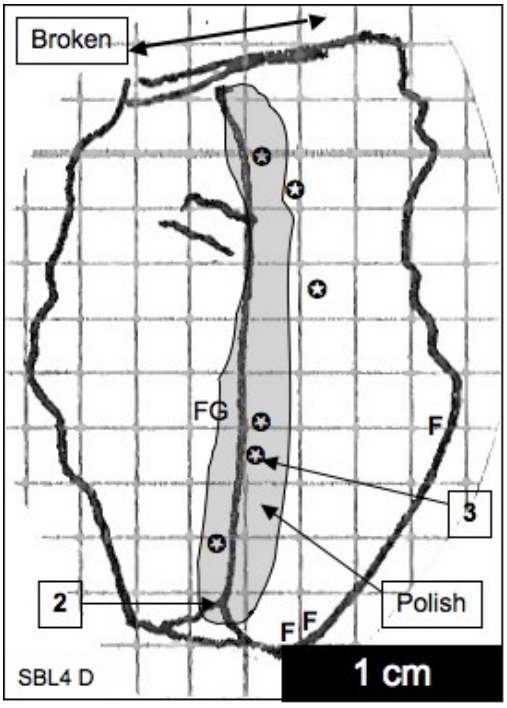


Figure D.17: Distribution map for SBL4, dorsal side. Note that the resin is located in the polish area. For key see Figure D.1.



Figure D.18: 1= Micrograph of bluish and yellowish fat on SBL4. Magnification 50.

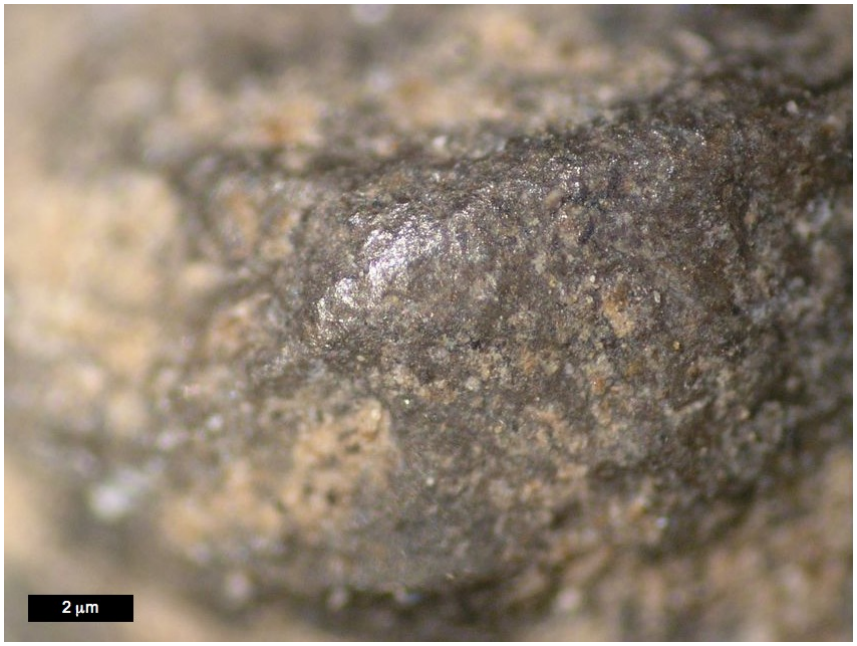


Figure D.19: 2= Micrograph of polish on SBL4. Magnification 50.



Figure D.20: 3= Micrograph of red ochre on SBL4. Magnification 100.

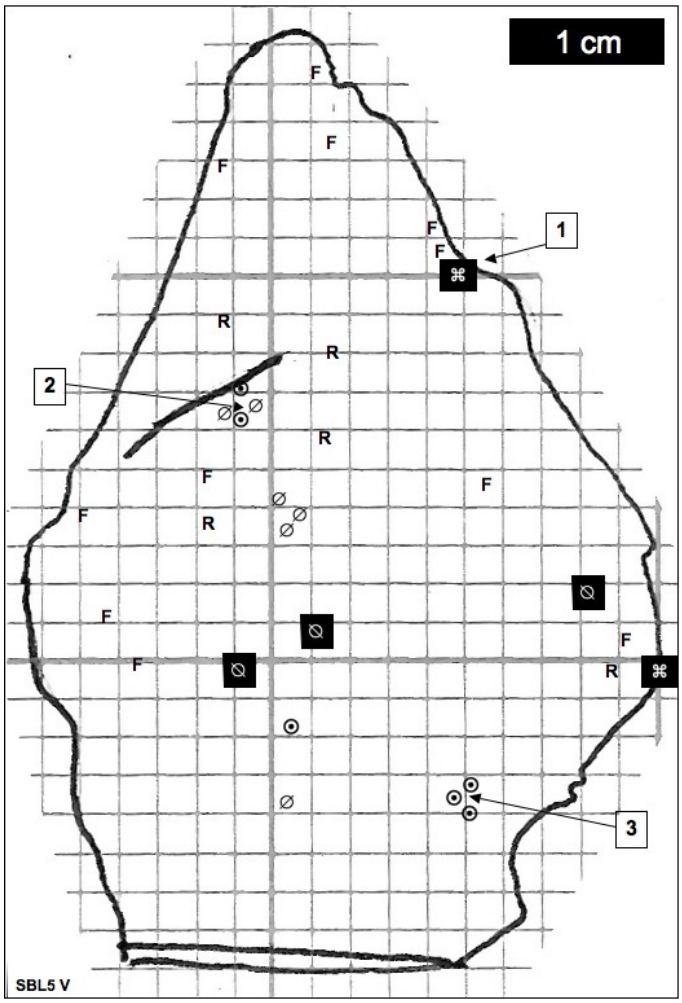


Figure D.21: Distribution map for SBL5, ventral side. For key see Figure D.1.

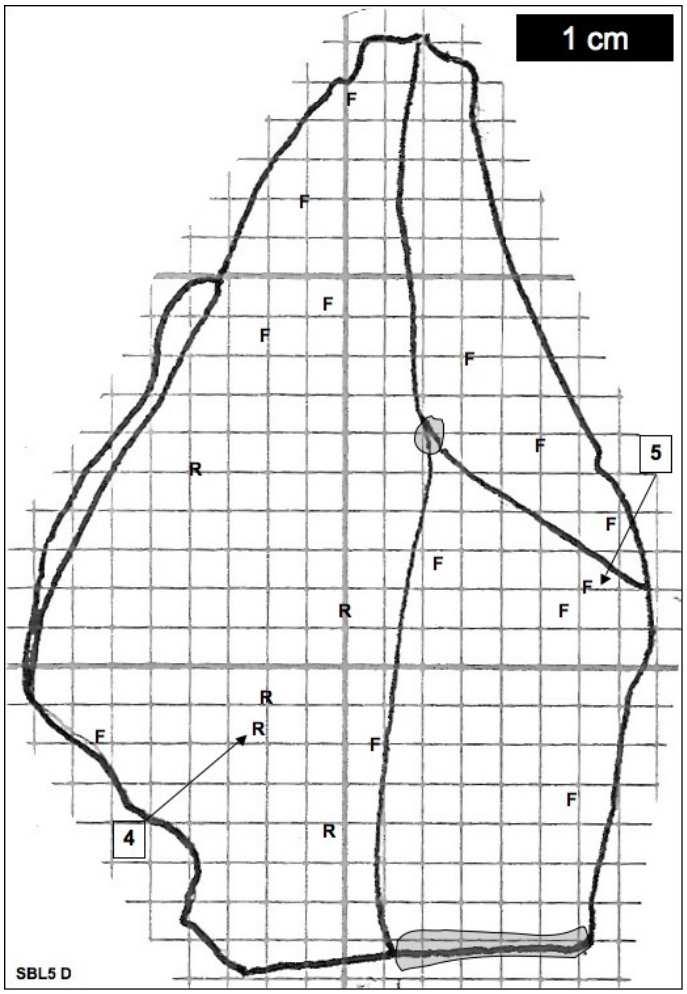


Figure D.22: Distribution map for SBL5, dorsal side. For key see Figure D.1.

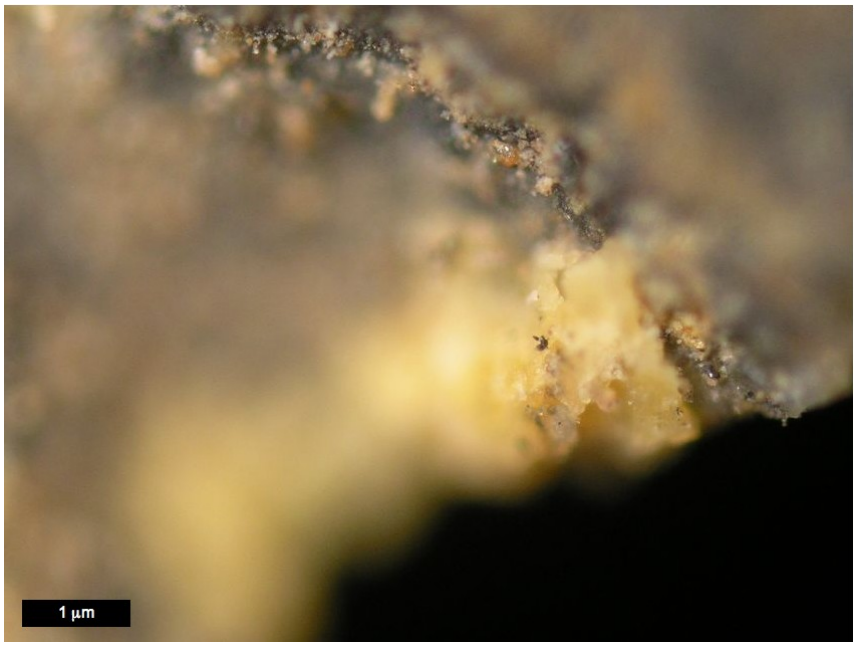


Figure D.23: 1= Micrograph of yellow burnt bone on SBL5. Magnification 100.



Figure D.24: 2= Micrograph of resin/plant tissue on SBL5. Magnification 50.

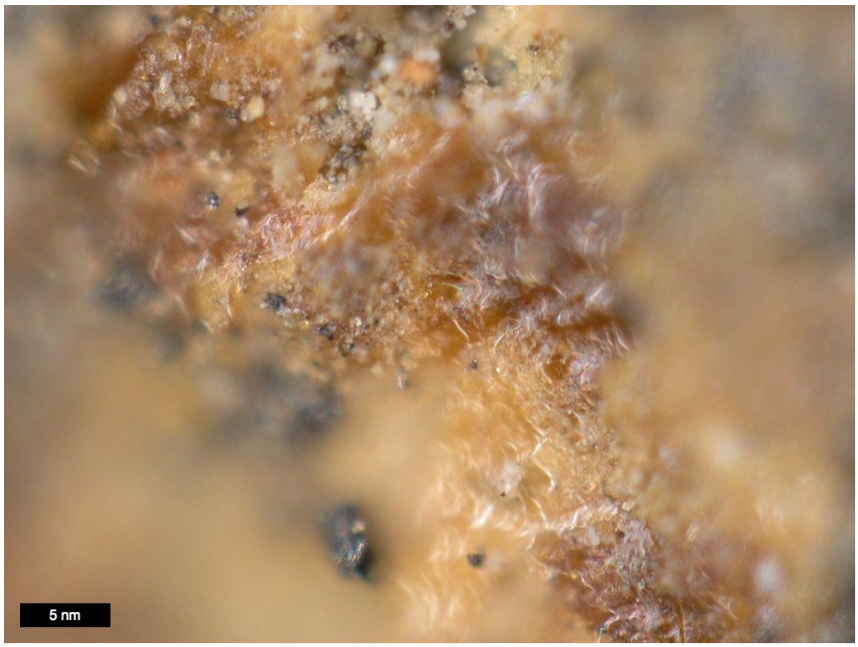


Figure D.25: 3= Micrograph of plant resin on SBL5. Magnification 200.

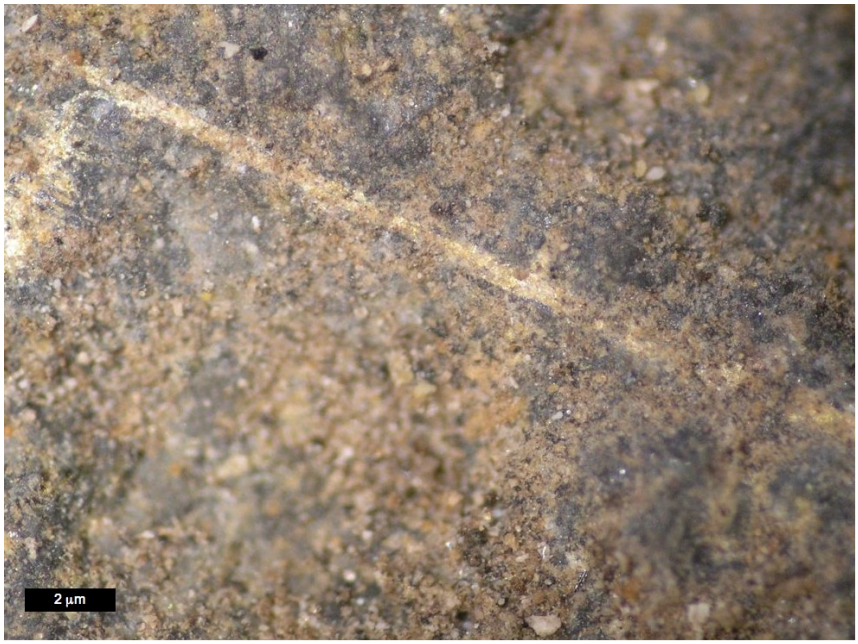


Figure D.26: 4= Micrograph of a rootlet on SBL5. Magnification 50.

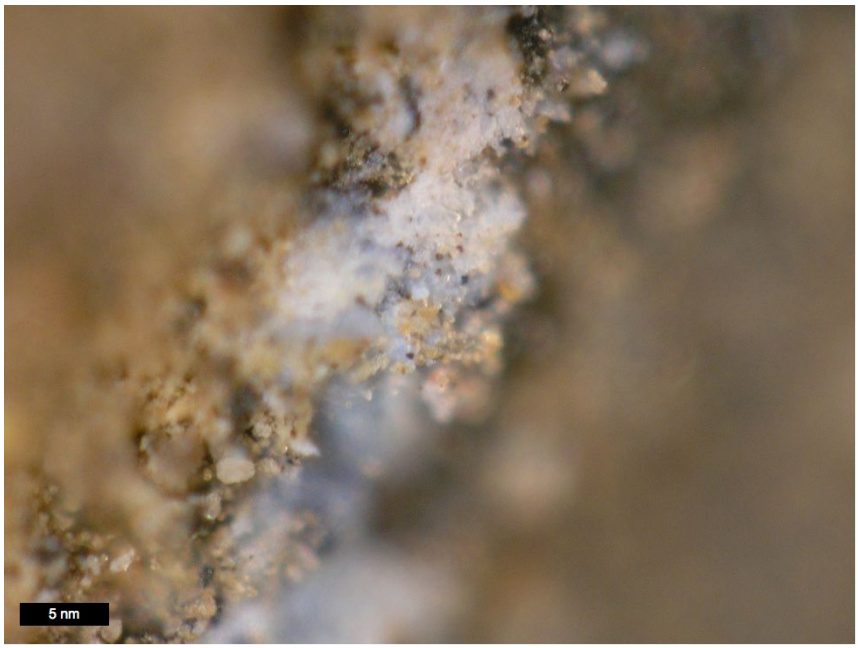


Figure D.27: 5= Micrograph of bluish fat on SBL5. Magnification 200.

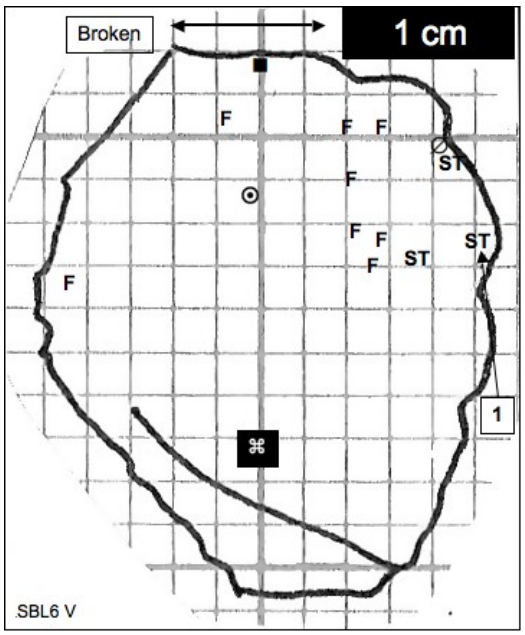


Figure D.28: Distribution map for SBL6, ventral side. For key see Figure D.1.

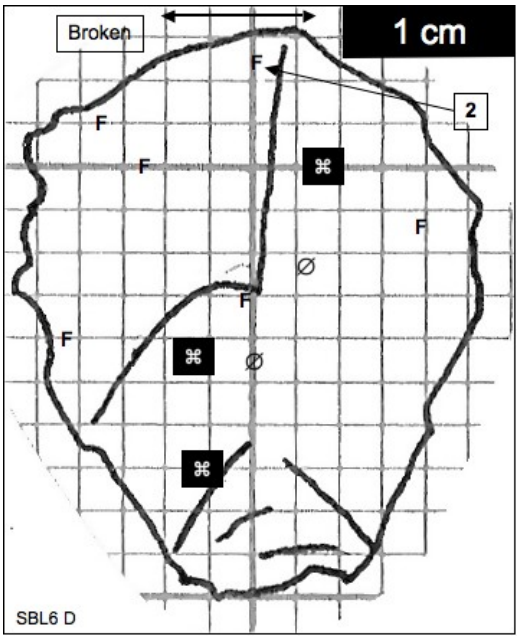


Figure D.29: Distribution map for SBL6, dorsal side. For key see Figure D.1.

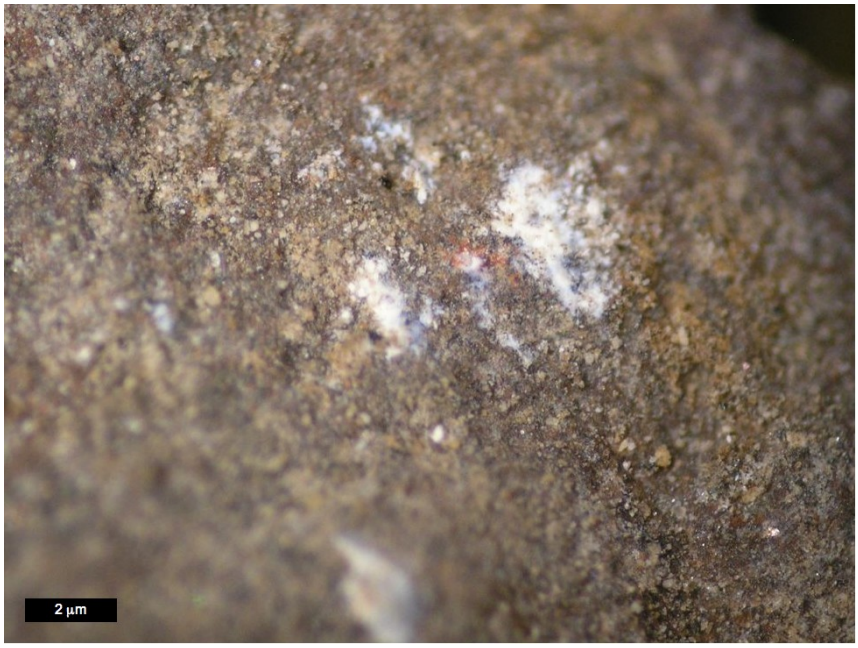


Figure D.30: 1= Micrograph of white/bluish fat on SBL6. Magnification 50.

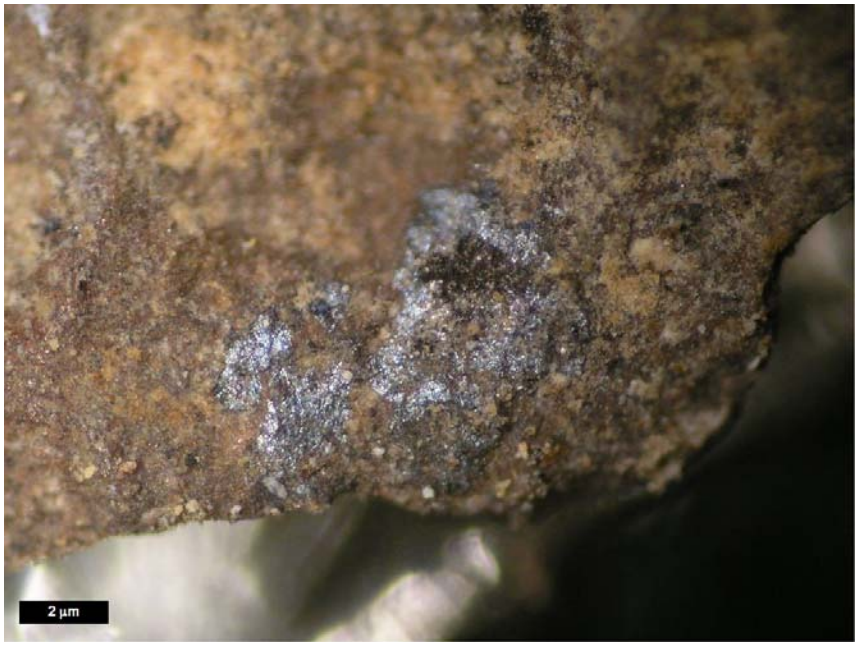


Figure D.31: 2= Micrograph of an unidentifiable stain on SBL6. Magnification 50.

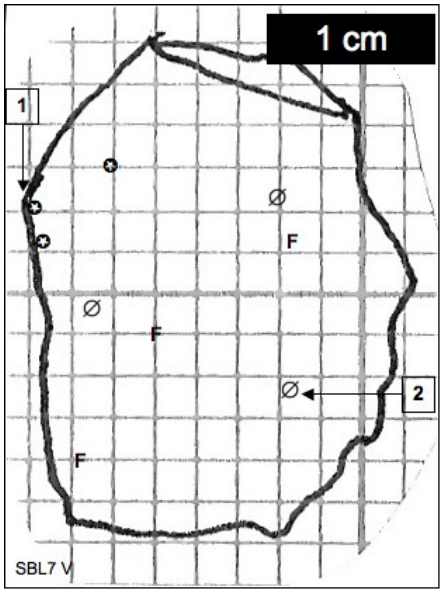


Figure D.32: Distribution map for SBL7, ventral side. For key see Figure D.1.

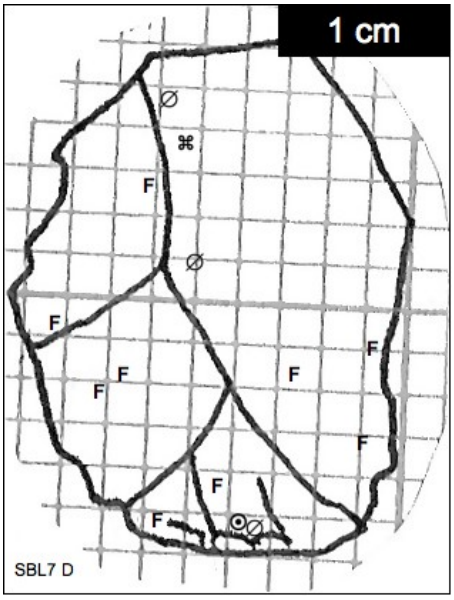


Figure D.33: Distribution map for SBL7, dorsal side. Not the random distribution of fat. For key see Figure D.1.

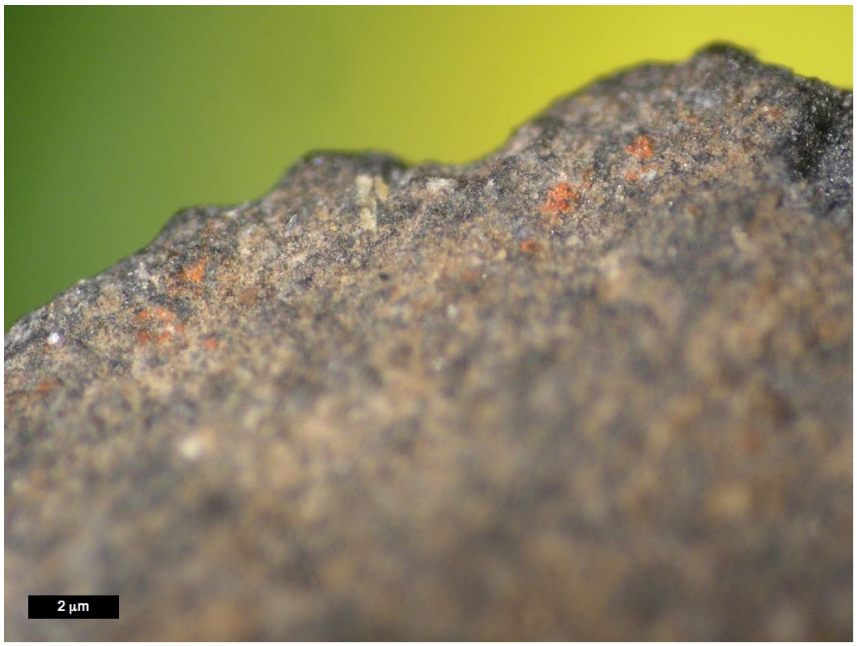


Figure D.34: 1= Micrograph of ochre spots on SBL7. Magnification 50.

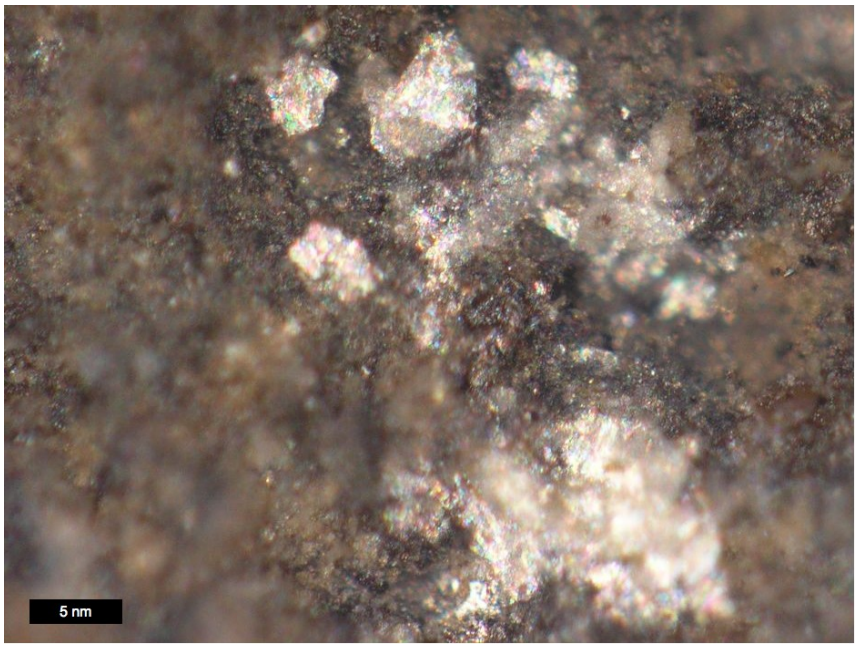


Figure D.35: 2= Micrograph of reflective plant remains on SBL7. Magnification 200.

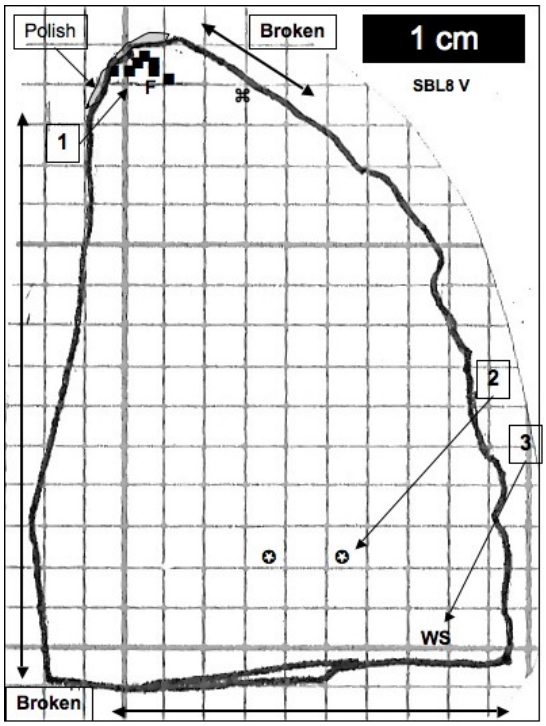


Figure D.36: Distribution map for SBL8, ventral side. For key see Figure D.1.

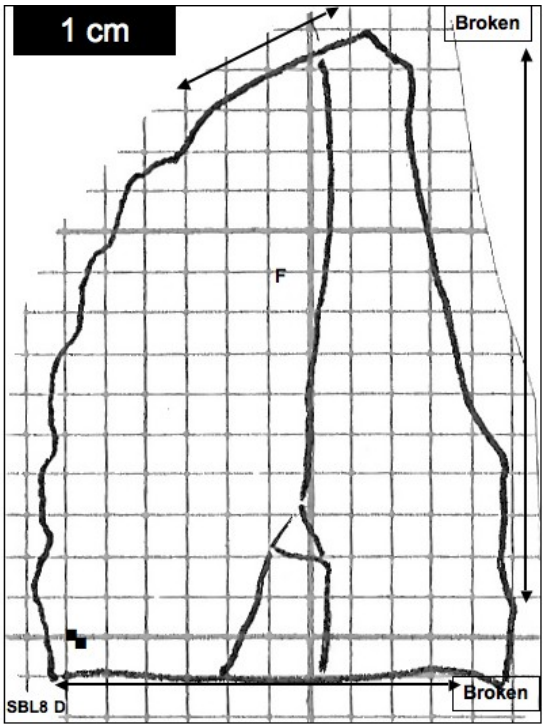


Figure D.37: Distribution map for SBL8, dorsal side. For key see Figure D.1.

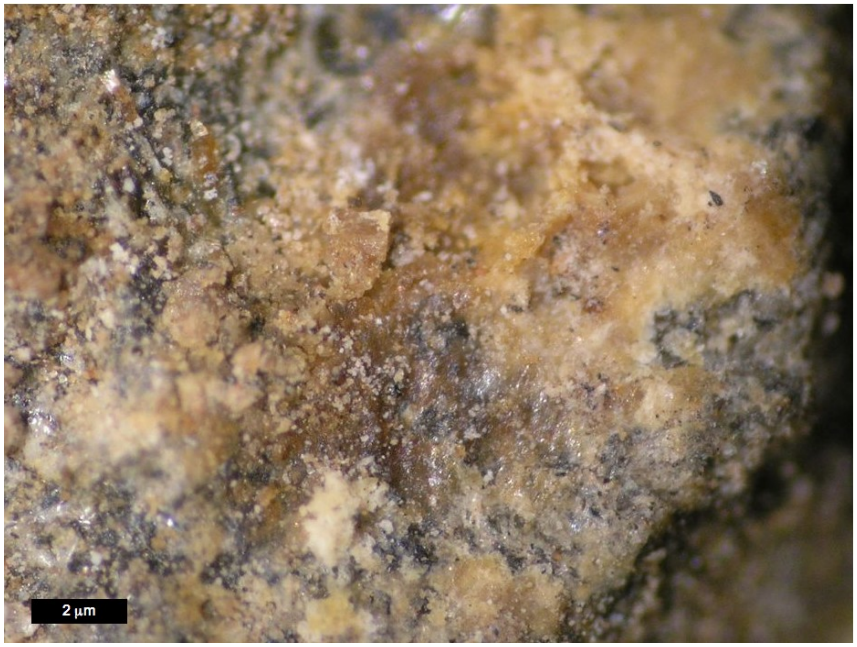


Figure D.38: 1= Micrograph of fatty animal tissue on SBL8. Magnification 50.



Figure D.39: 2= Micrograph of an ochre spot on SBL8. Magnification 50.



Figure D.40: 3= Micrograph of a "white starchy deposit" on SBL8. Magnification 50.

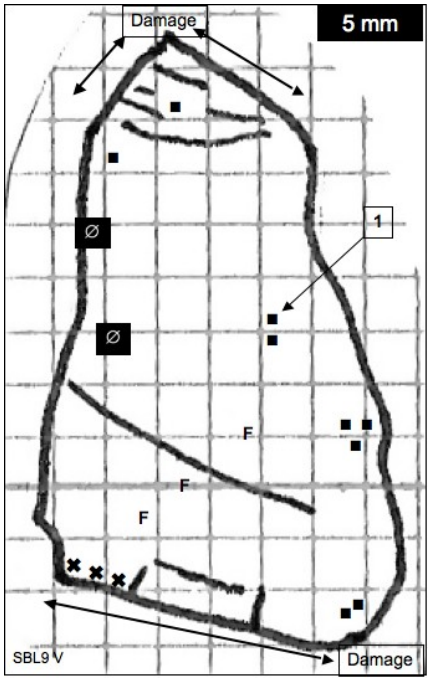


Figure D.41: Distribution map for SBL9, ventral side. Note the abundant animal type residues. For key see Figure D.1.

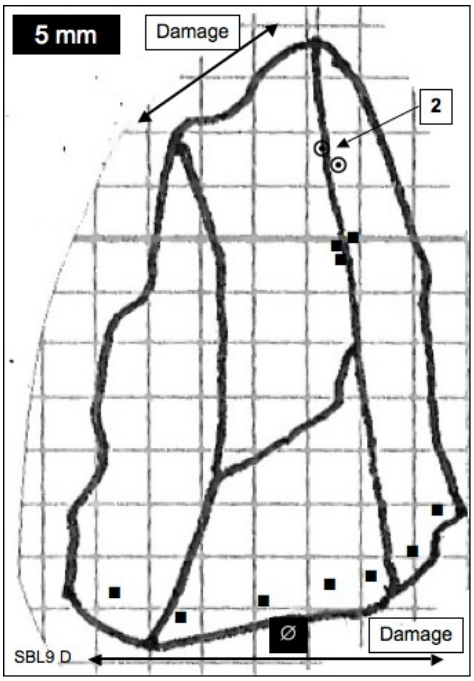


Figure D.42: Distribution map for SBL9, dorsal side. Note the abundant animal type residues. For key see Figure D.1.

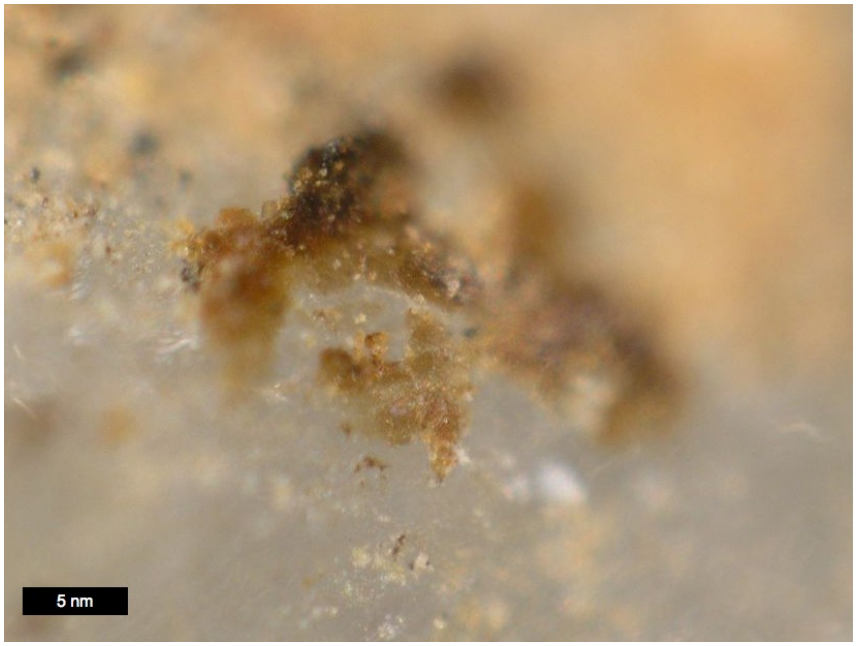


Figure D.43: 1= Micrograph of muscle tissue on SBL9. Magnification 200.

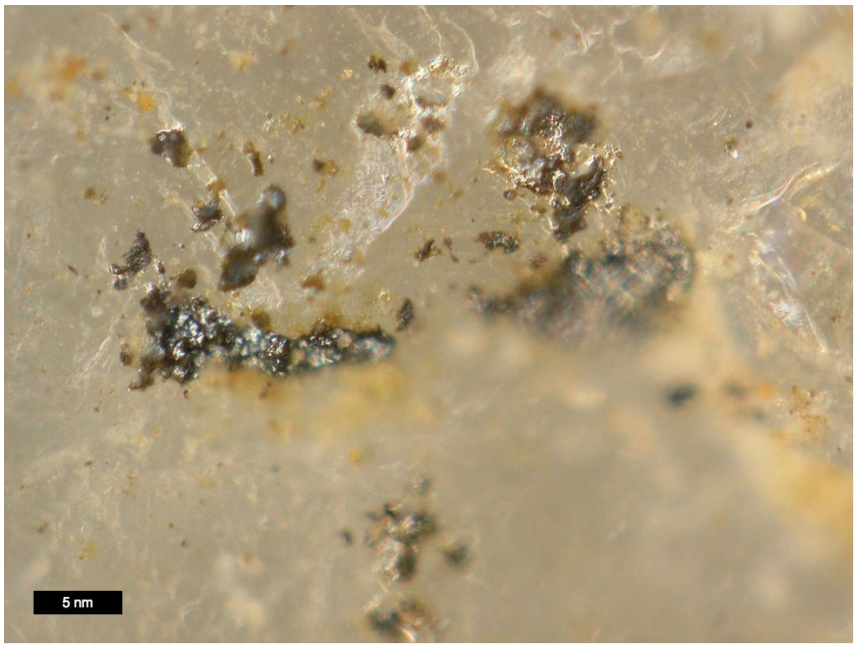


Figure D.44: 1= Micrograph of resin drops on SBL9. Magnification 200.

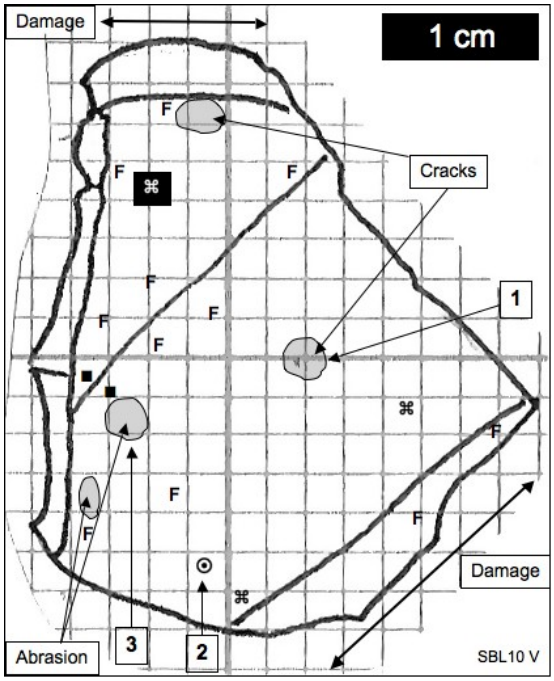


Figure D.45: Distribution map for SBL10, ventral side. Note that animal type residues are present in suite and mainly located around the edges. For key see Figure D.1.

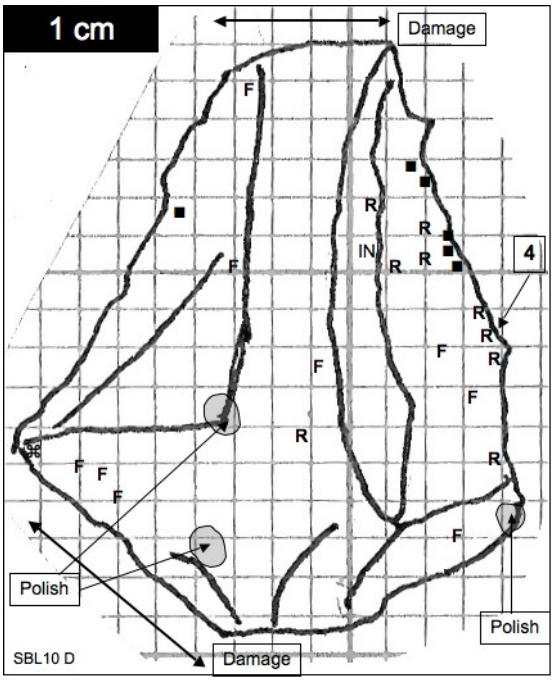


Figure D.46: Distribution map for SBL1, dorsal side. Note that animal type residues are present in suite and mainly located around the edges. For key see Figure D.1.



Figure D.47: 1= Micrograph of the cracked stone surface on SBL10. Magnification 50.

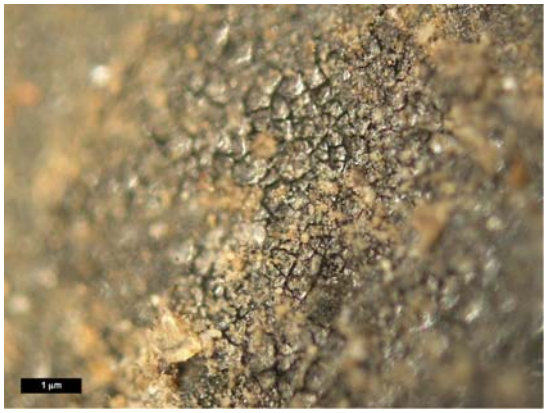


Figure D.48: 1= Micrograph of the cracked stone surface on SBL10. Magnification 100.



Figure D.49: 1= Micrograph of the cracked stone surface on SBL10. Magnification 200.



Figure D.50: 2= Micrograph of resin on SBL10. Note the little crack in the surface, indicated by the arrow. Magnification 100.



Figure D.51: 3= Micrograph of reflective abrasion on SBL10. Magnification 50.

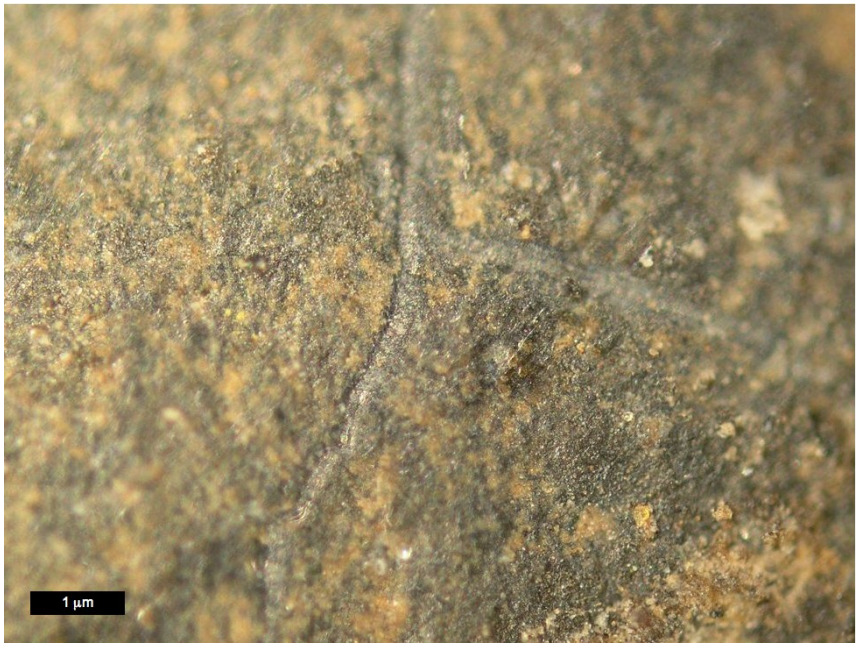


Figure D.52: 4= Micrograph of a rootlet negative on SBL10. Magnification 100.

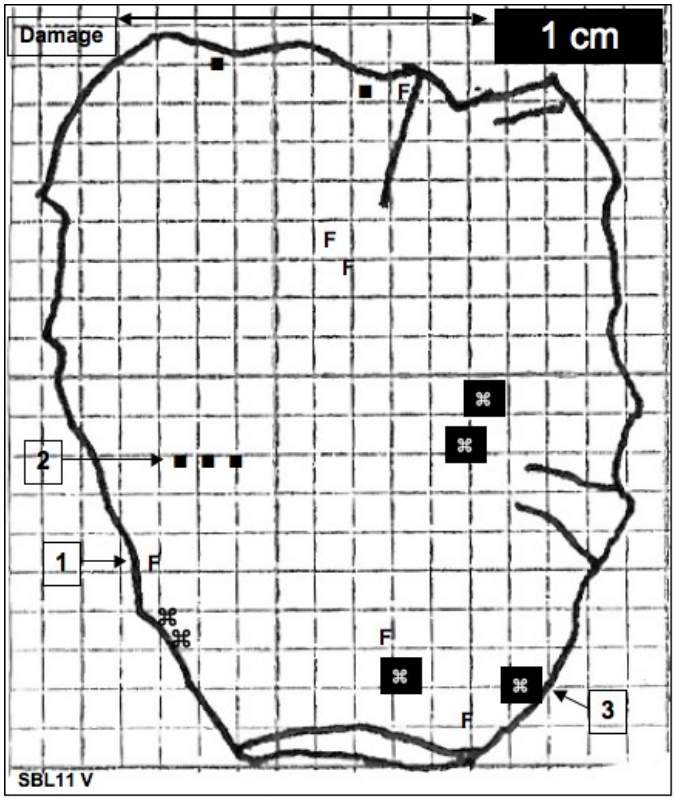


Figure D.53: Distribution map for SBL11, ventral side. For key see Figure D.1.

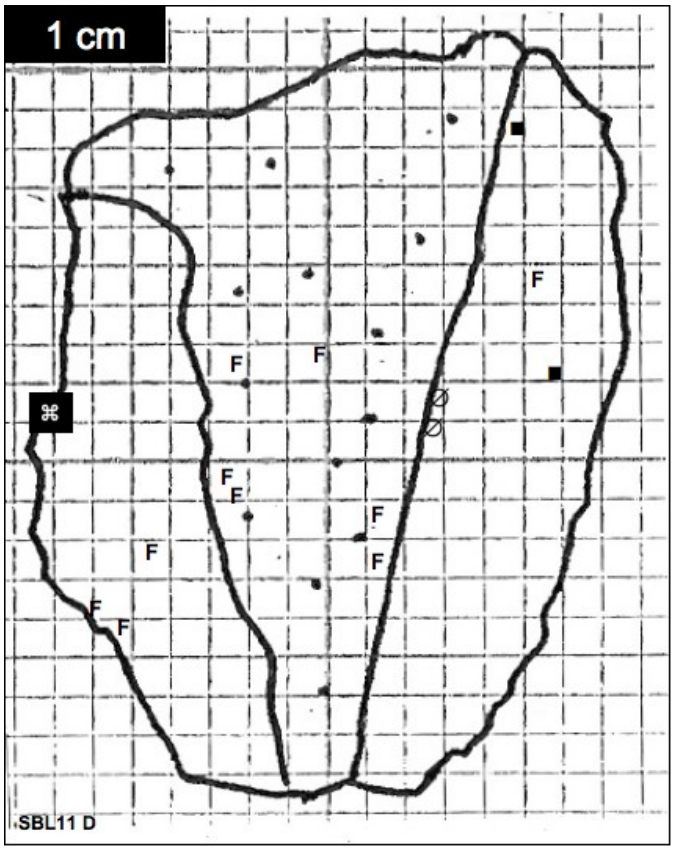


Figure D.54: Distribution map for SBL11, dorsal side. The fat deposits have an arbitrary distribution. For key see Figure D.1.

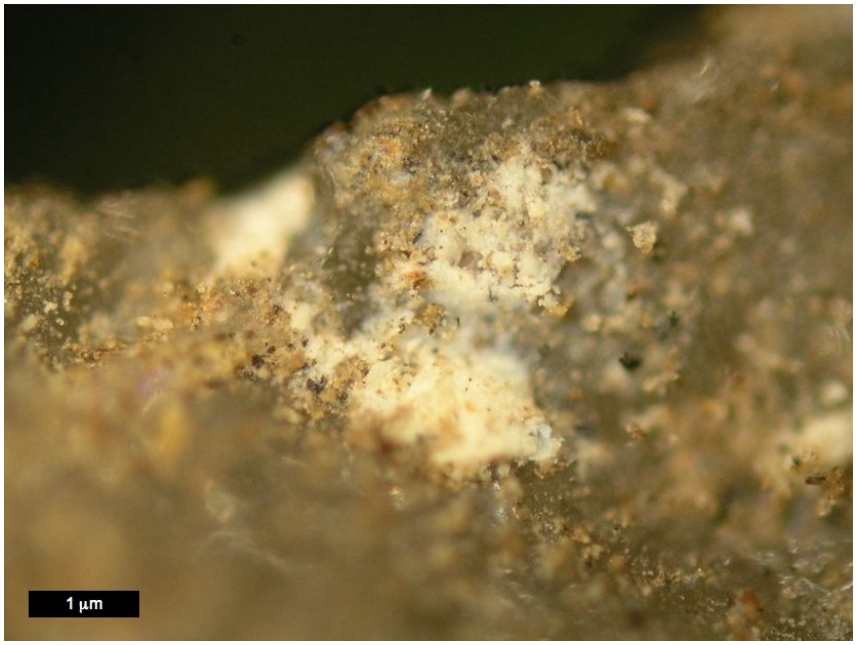


Figure D.55: 1= Micrograph of white/bluish fat on SBL11. Magnification 100.

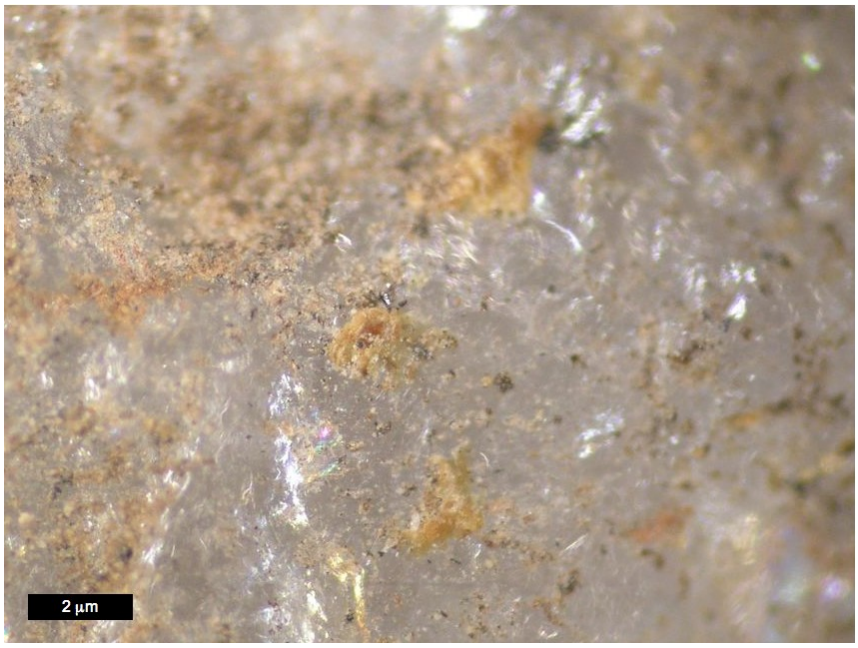


Figure D.56: 2= Micrograph of muscle tissue on SBL11. Magnification 50.

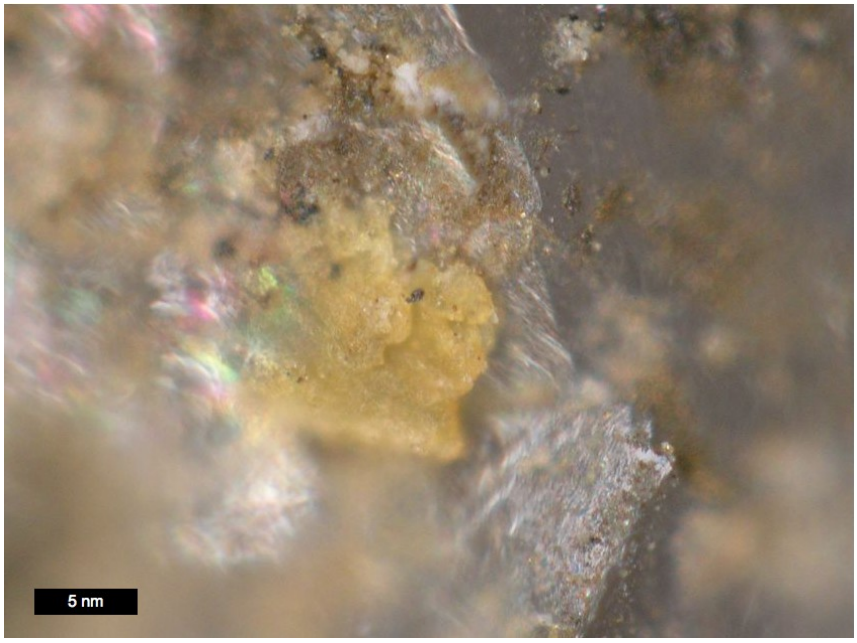


Figure D.57: 3= Micrograph of burnt fatty bone on SBL11. Magnification 200.

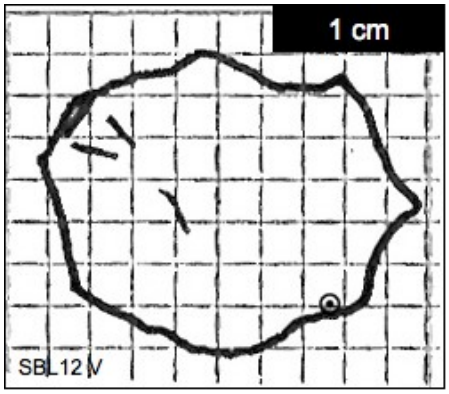


Figure D.58: Distribution map for SBL12, ventral side. For key see Figure D.1.

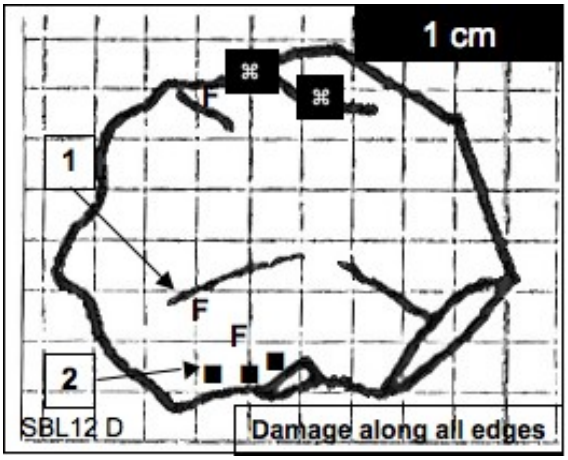


Figure D.59: Distribution map for SBL12, dorsal side. For key see Figure D.1.



Figure D.60: 1= Micrograph of white/bluish fat on SBL12. Note that the fat is located under a sediment patch. Magnification 100.

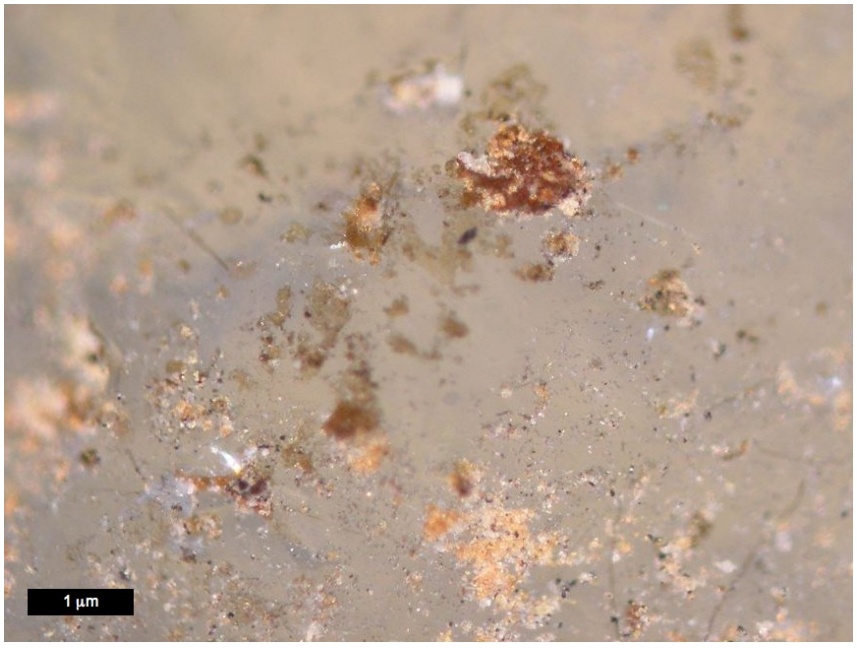


Figure D.61: 1= Micrograph of muscle tissue on SBL12. Because the background is reflective, the muscle tissue appears darker. Magnification 100.

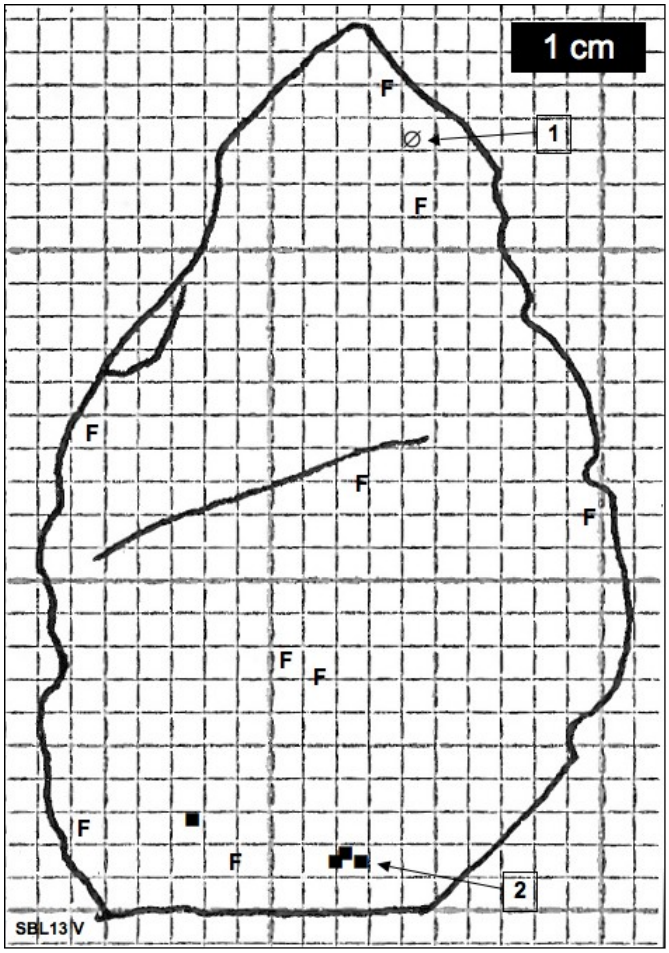


Figure D.62: Distribution map for SBL13, ventral side. For key see Figure D.1.

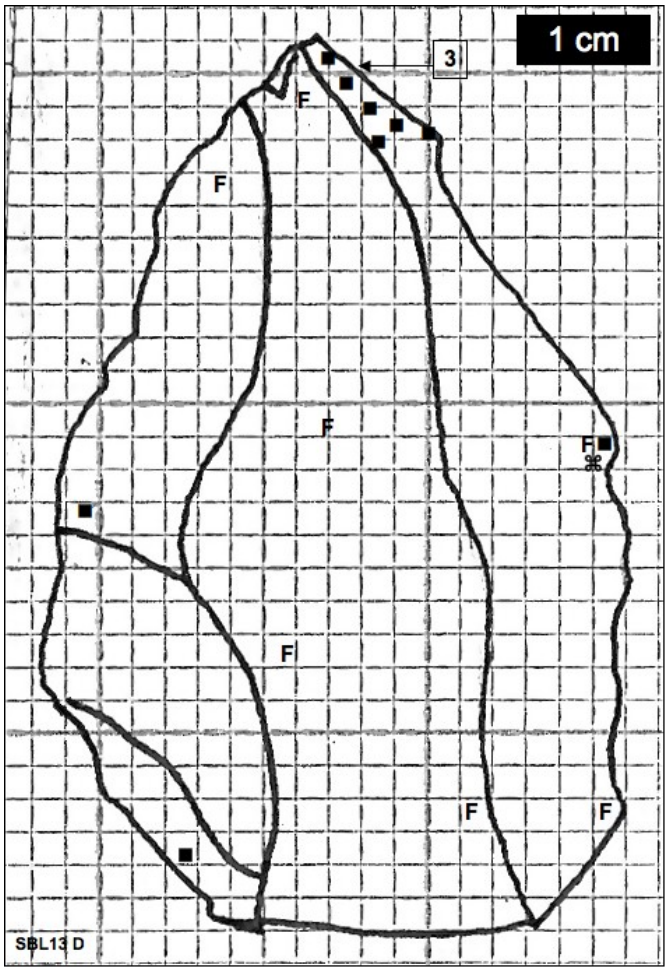


Figure D.63: Distribution map for SBL13, dorsal side. For key see Figure D.1.

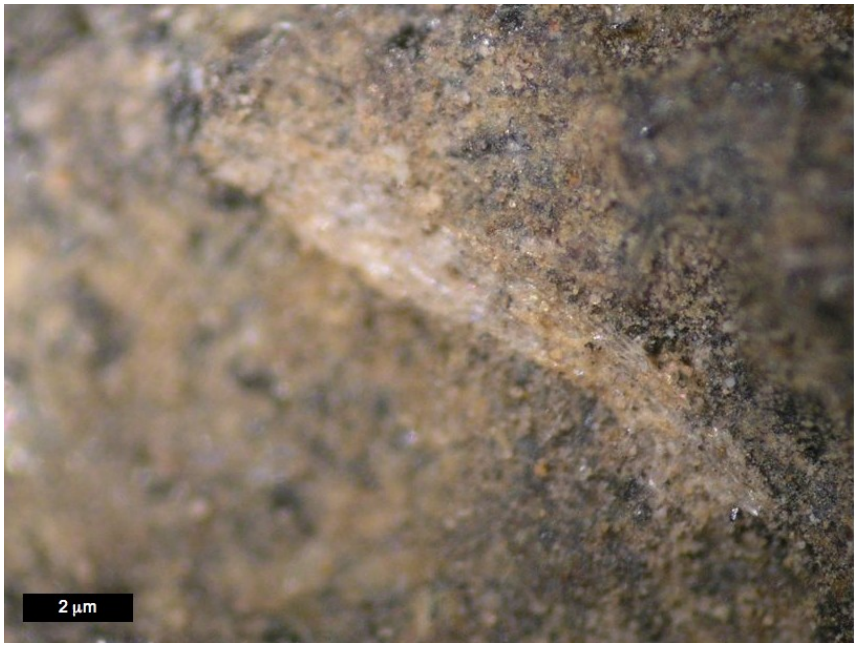


Figure D.64: 1= Micrograph of plant tissue on SBL13. Magnification 50.

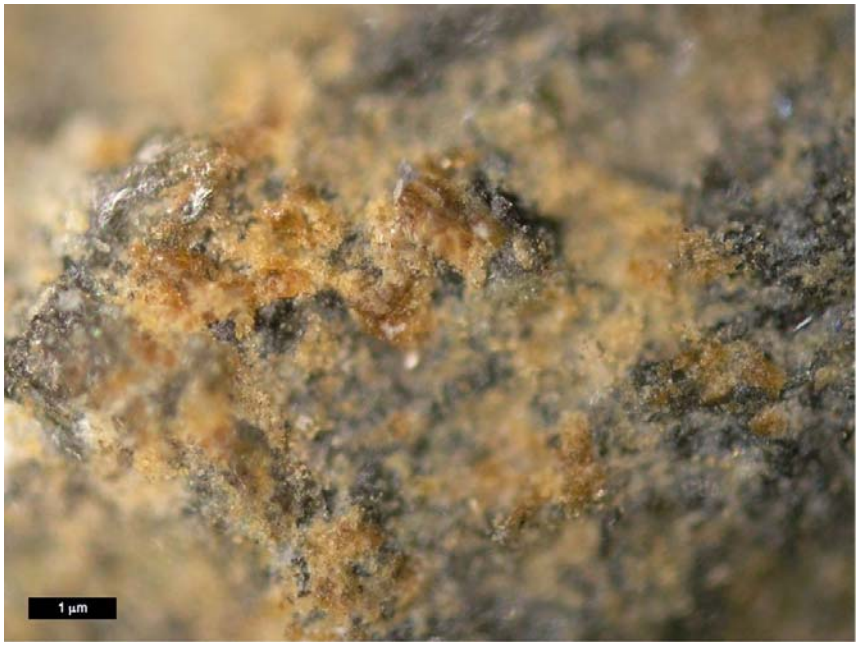


Figure D.65: 2= Micrograph of brown muscle tissue on SBL13. Magnification 100.



Figure D.66: 3= Micrograph of brown muscle tissue on SBL13. Magnification 50.

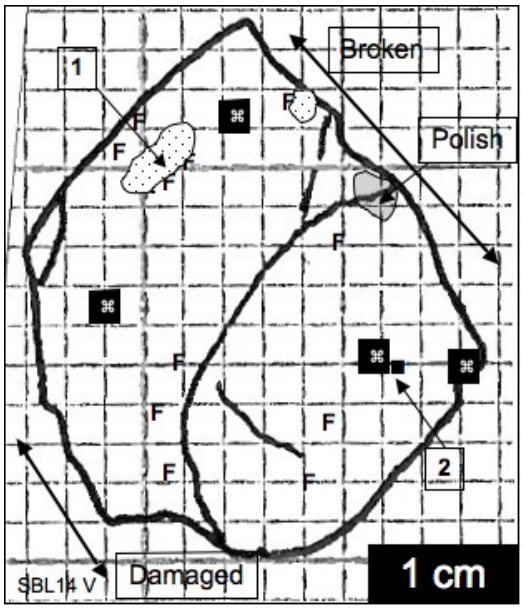


Figure D.67: Distribution map for SBL14, ventral side. For key see Figure D.1.

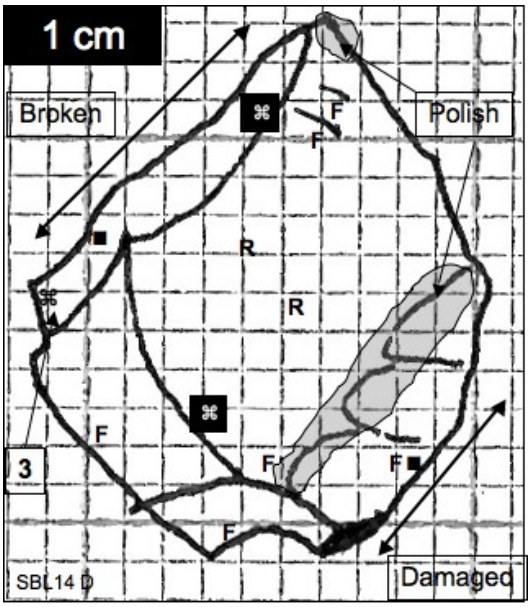


Figure D.68: Distribution map for SBL14, dorsal side. For key see Figure D.1.

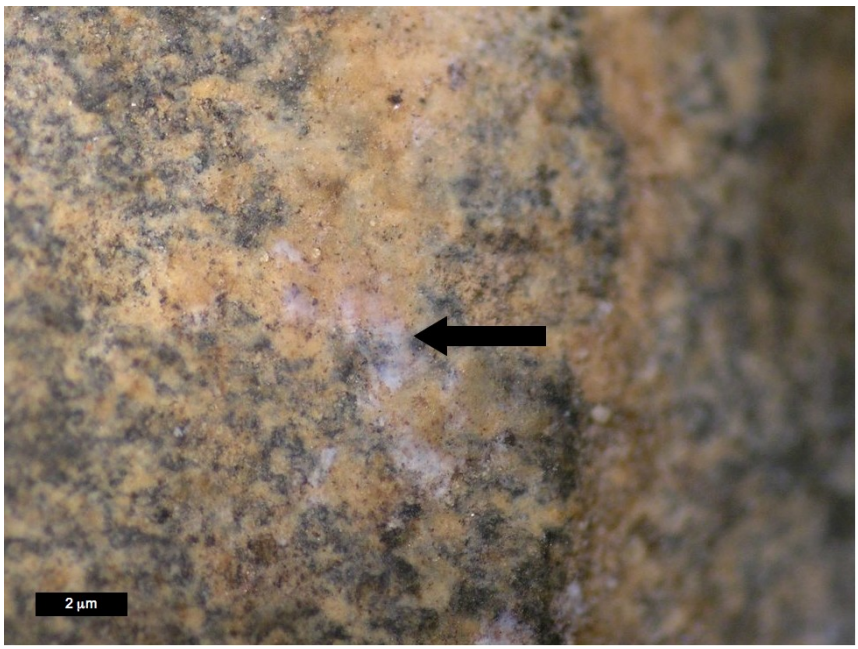


Figure D.69:1=Micrograph of fat under sediment (indicated with the arrow) on SBL14. Magnification 50.



Figure D.70: 2= Micrograph of burnt bone on SBL14. Magnification 50.

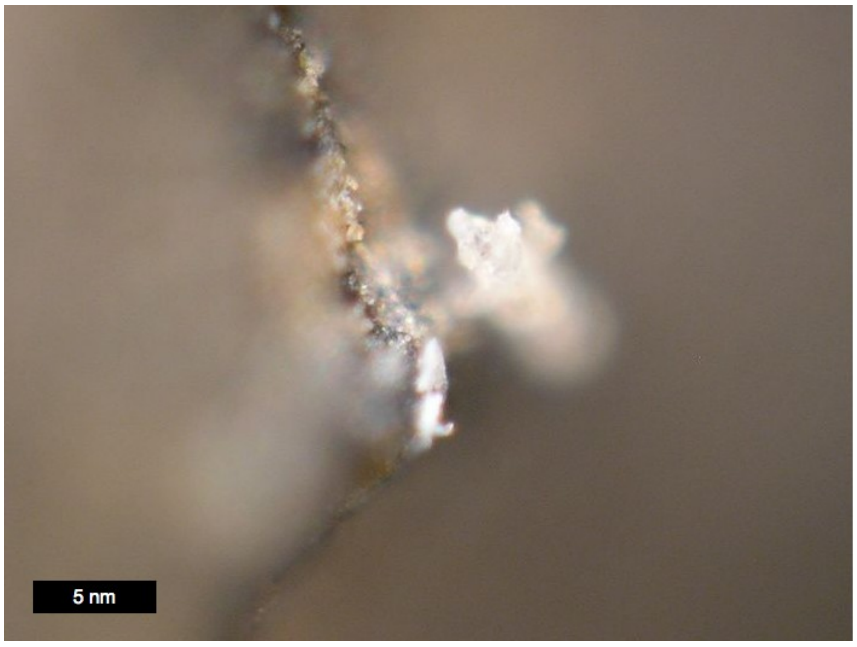


Figure D.71: 3= Micrograph of connective tissue on SBL14. Note that the tissue reflective. Magnification 200.

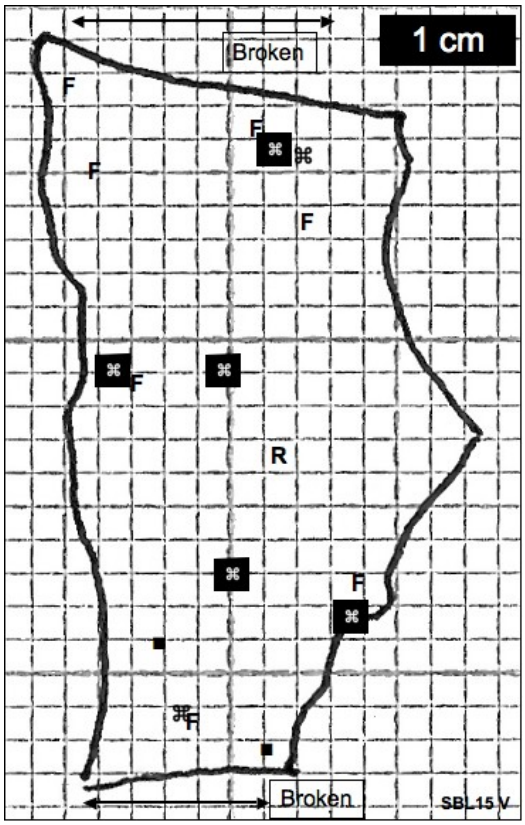


Figure D.72: Distribution map for SBL15, ventral side. Note the random distribution the remains. For key see Figure D.1.

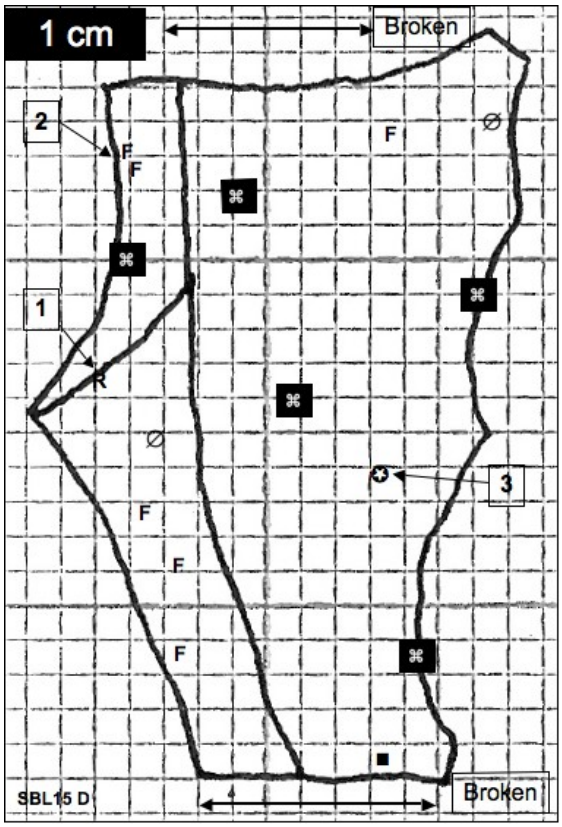


Figure D.73: Distribution map for SBL15, dorsal side. Note the random distribution the remains. For key see Figure D.1.



Figure D.74: 1= Micrograph of a plant tissue on SBL15. Note the rigid and linear organisation of the plant tissue. Magnification 50.



Figure D.75: 2= Micrograph of fat on top of a sediment deposit on SBL15. The edges of the fat deposit are bluish. Magnification 200.



Figure D.76: 3= Micrograph of red ochre on SBL15. Magnification 200.

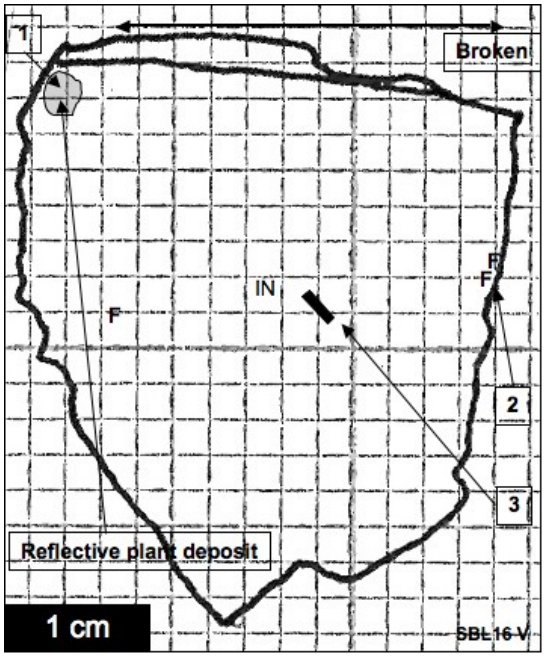


Figure D.77: Distribution map for SBL16, ventral side. For key see Figure D.1.

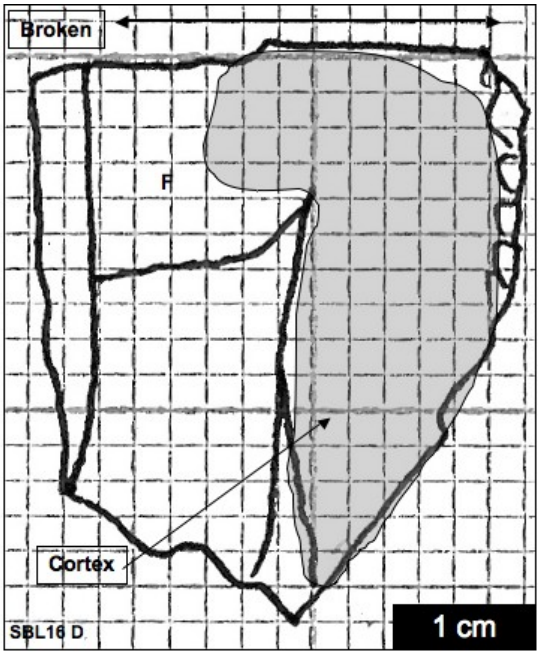


Figure D.78: Distribution map for SBL16, dorsal side. For key see Figure D.1.

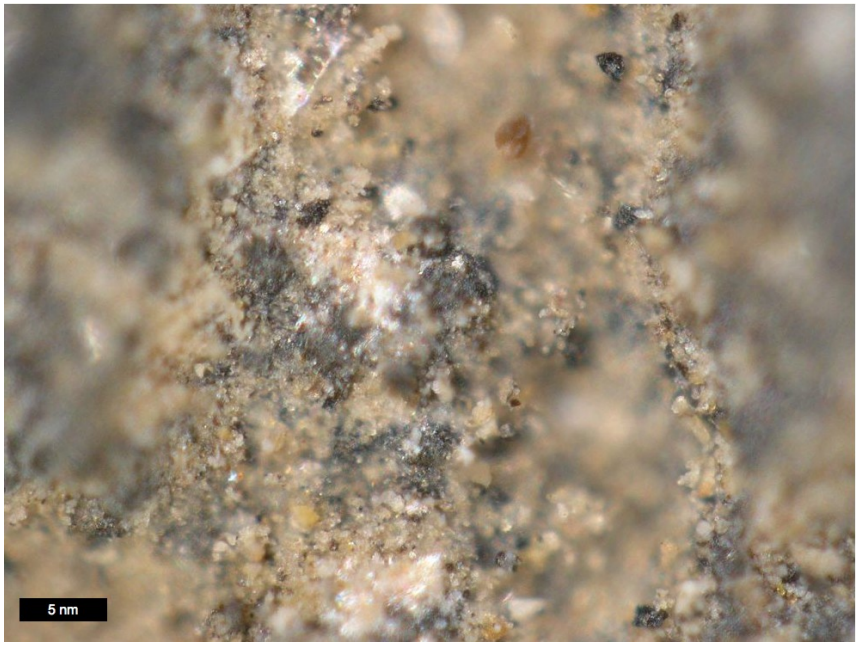


Figure D.79: 1= Micrograph of reflective plant tissue in sediment matrix on SBL16. Magnification 200.

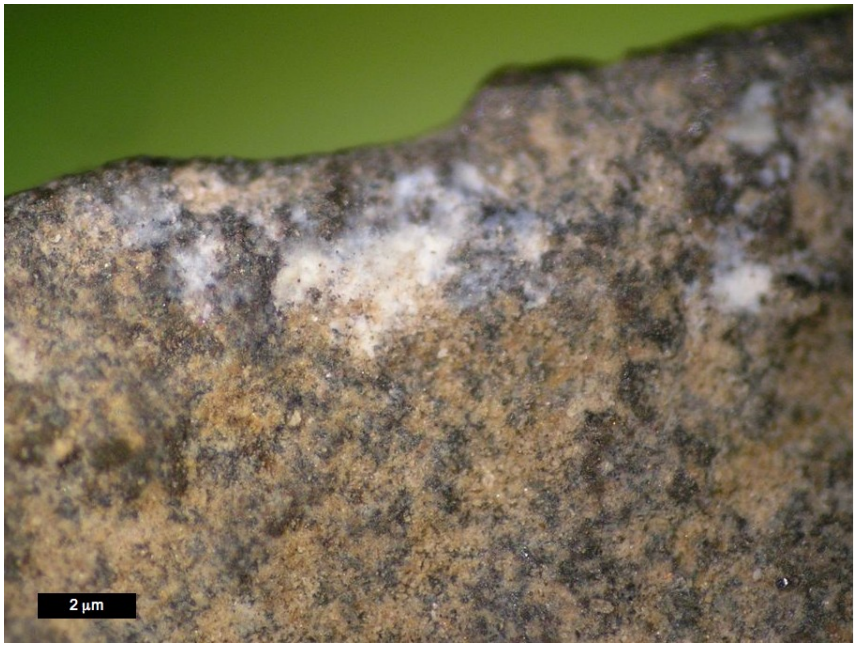


Figure D.80: 2= Micrograph of white/bluish fat under sediment on SBL16. Magnification 50.



Figure D.81: 3= Micrograph of a modern plant fibre on SBL16. The plant fibre is located on top of sediment. Magnification 100.

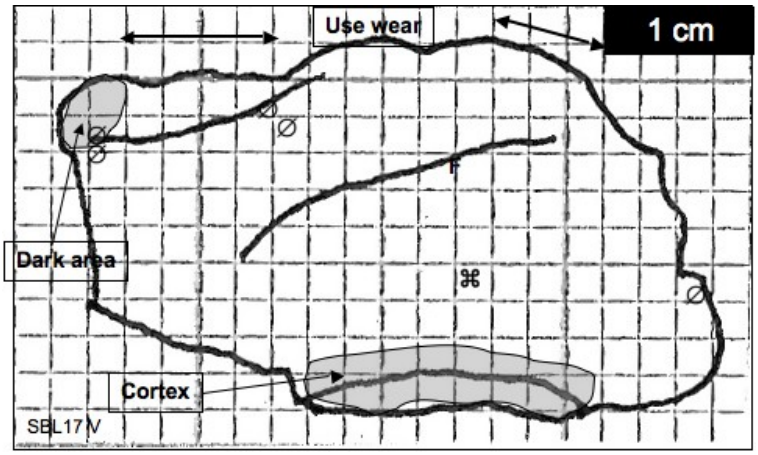


Figure D.82: Distribution map for SBL17, ventral side. For key see Figure D.1.

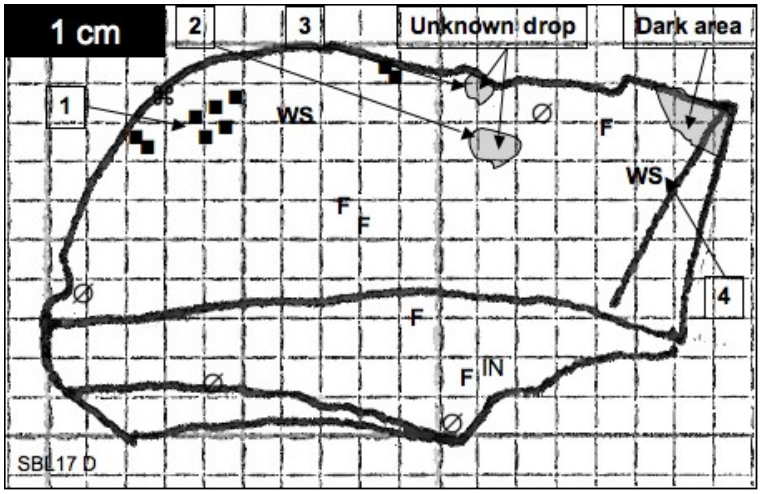


Figure D.83: Distribution map for SBL17, dorsal side. For key see Figure D.1.



Figure D.84: 1= Micrograph of animal tissue on SBL17. Magnification 50.

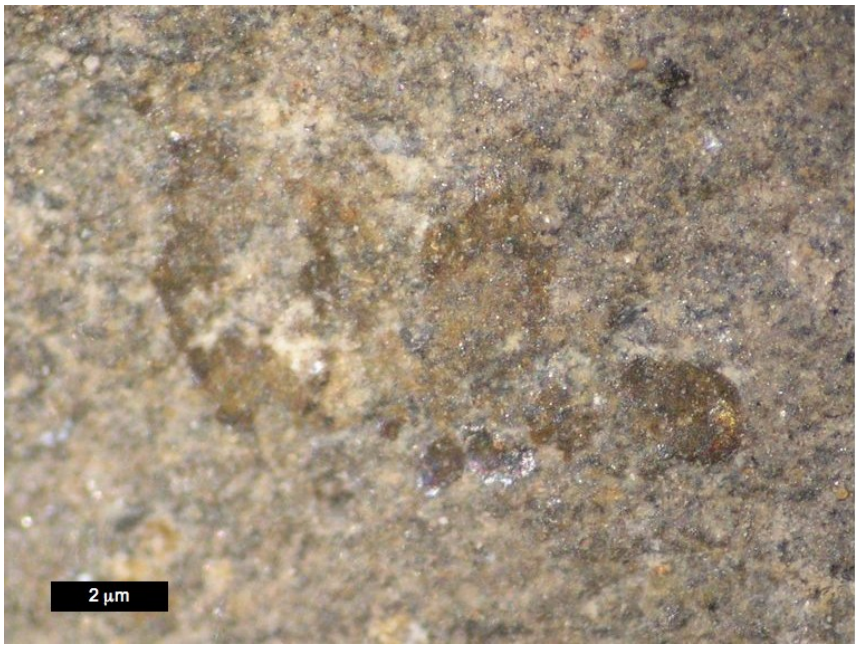


Figure D.85: 2= Micrograph of an unidentified stain or drop on SBL17. Magnification 50.

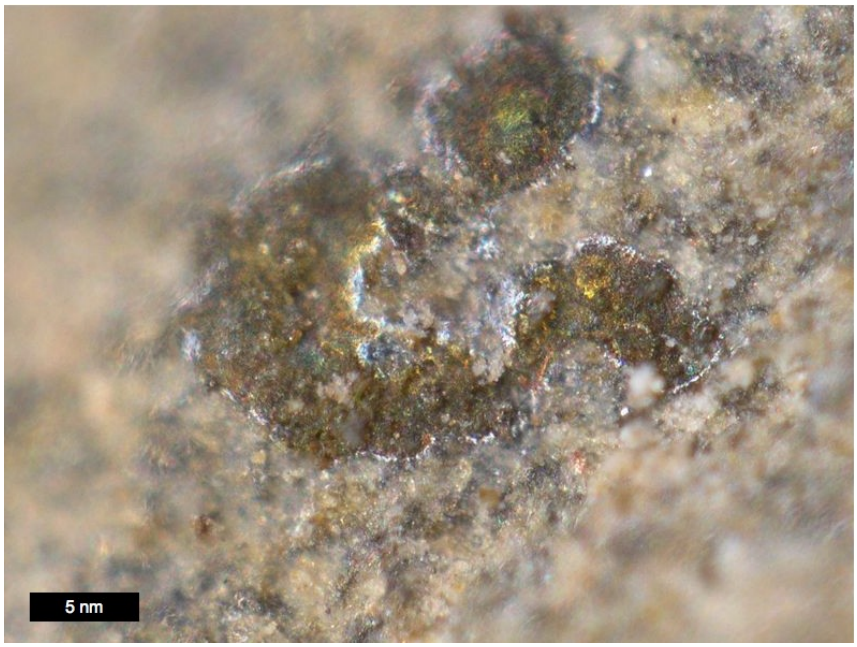


Figure D.86: 3= Micrograph of an unidentified stain or drop on SBL17. Magnification 200.

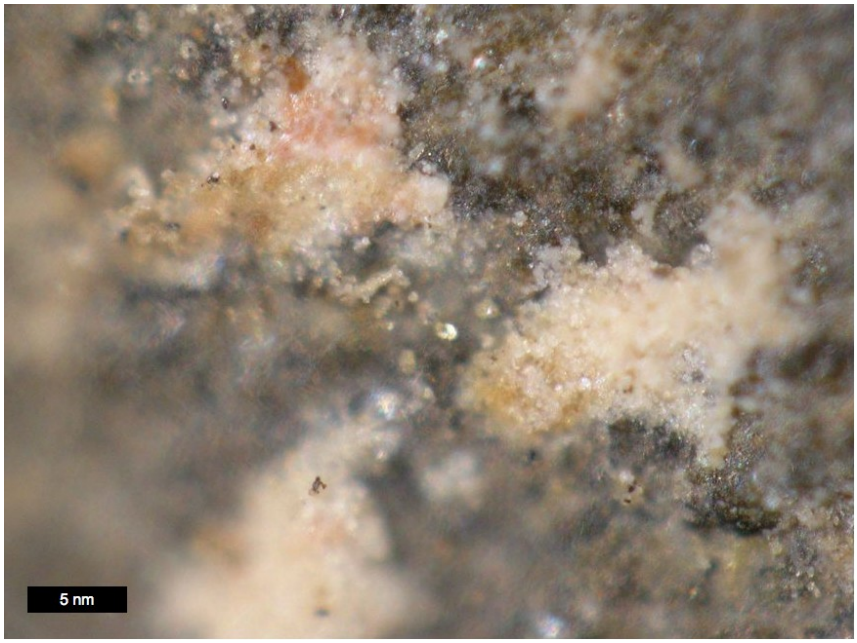


Figure D.87: 4= Micrograph of a "white starchy deposit" on SBL17. The deposit lays on top sediment. Magnification 200.

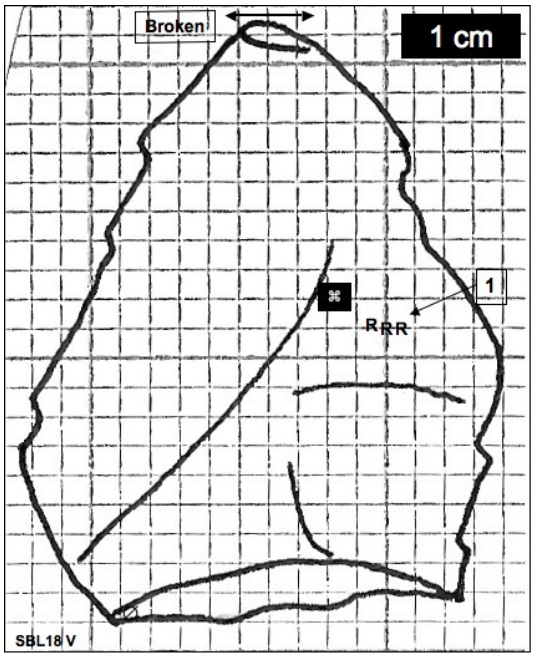


Figure D.88: Distribution map for SBL18, ventral side. For key see Figure D.1.

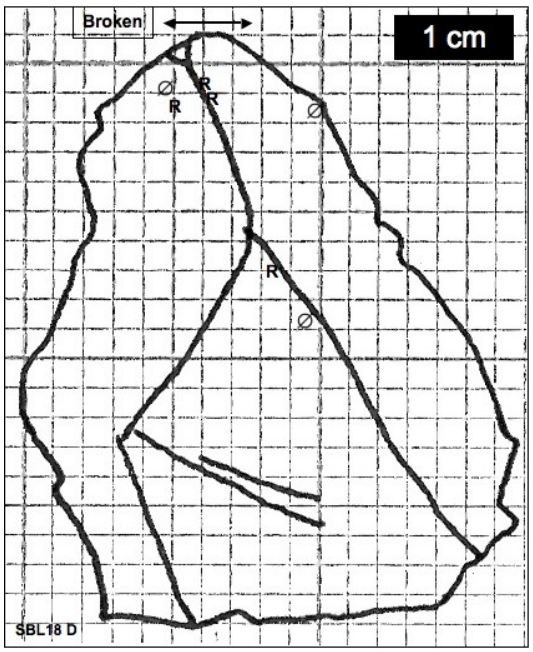


Figure D.89: Distribution map for SBL18, dorsal side. Because there are many rootlet remains on this artefact, the other plant tissue deposits are disregarded. For key see Figure D.1.

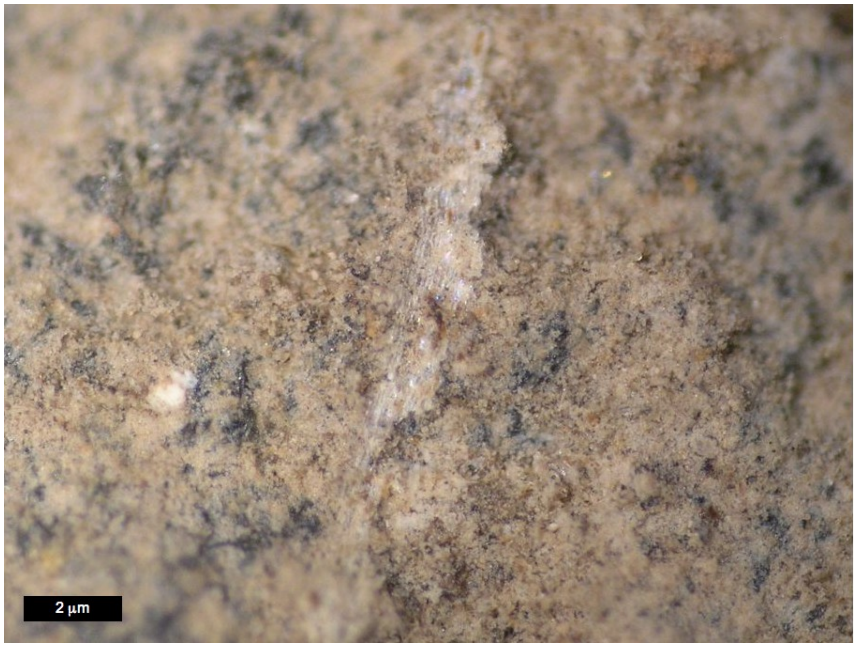


Figure D.90: 1= Micrograph of a rootlet on SBL18. Magnification 50.

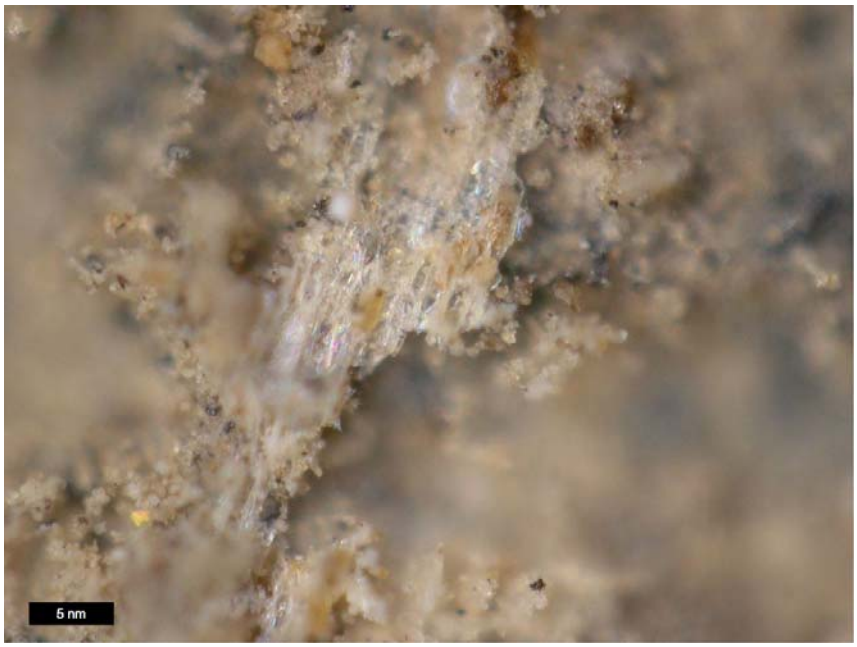


Figure D.91: 1= Micrograph of a rootlet on SBL18. Note the rigid cell structure of the plant tissue. Magnification 200.

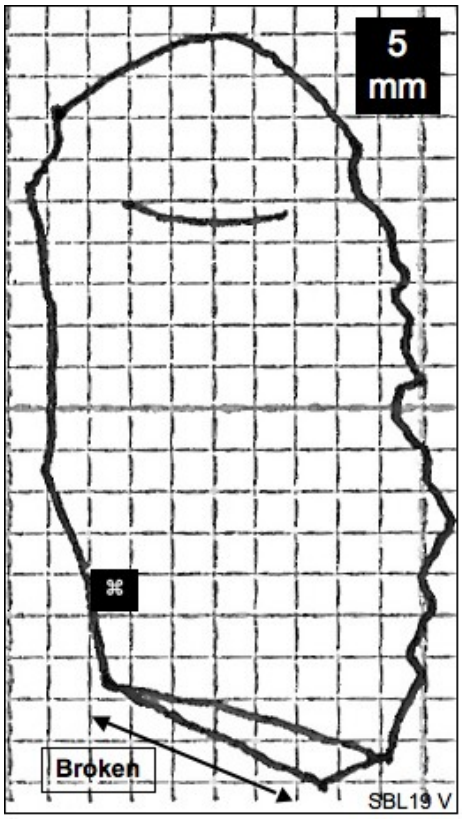


Figure D.92: Distribution map for SBL19, ventral side. For key see Figure D.1.

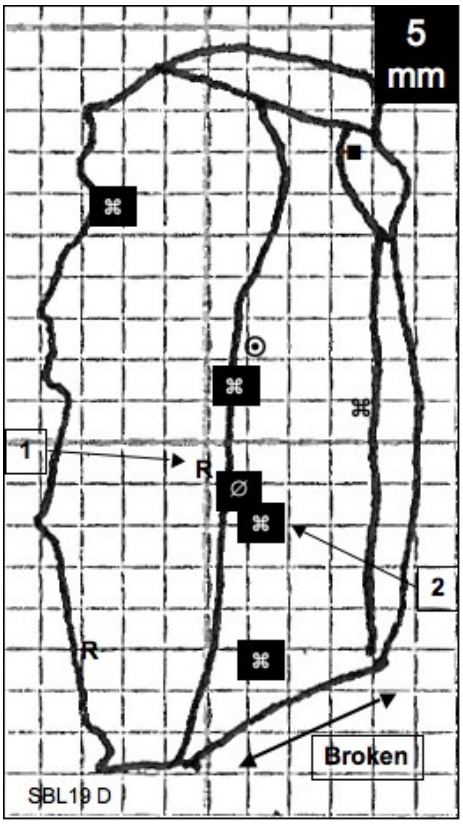


Figure D.93: Distribution map for SBL19, dorsal side. For key see Figure D.1.

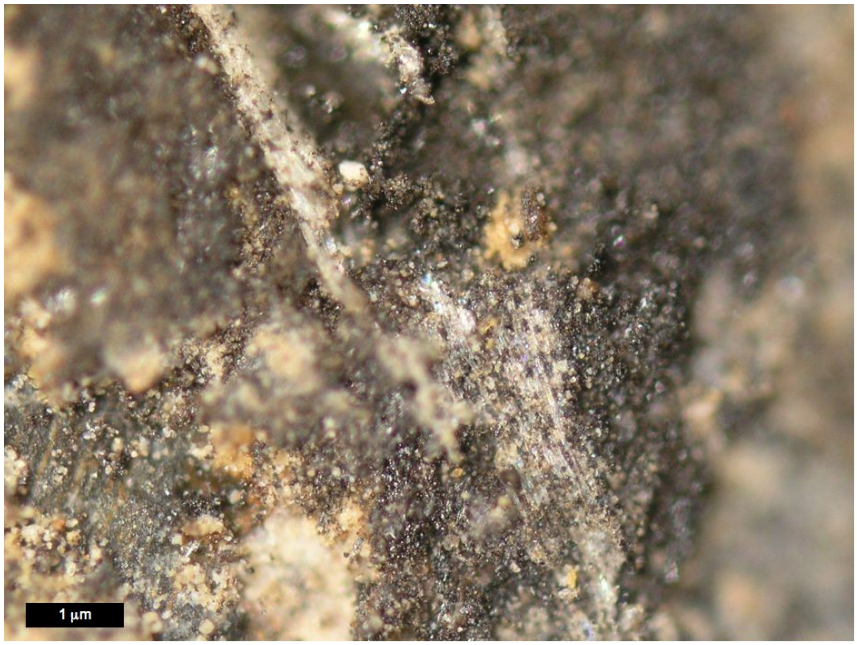


Figure D.94: 1= Micrograph of a rootlet on SBL19. These remains are similar to the rootlet remains on SBL18. Magnification 100.



Figure D.95: 2= Micrograph of burnt bone on SBL19. The deposit appears to be smeared on the surface. Magnification 50.

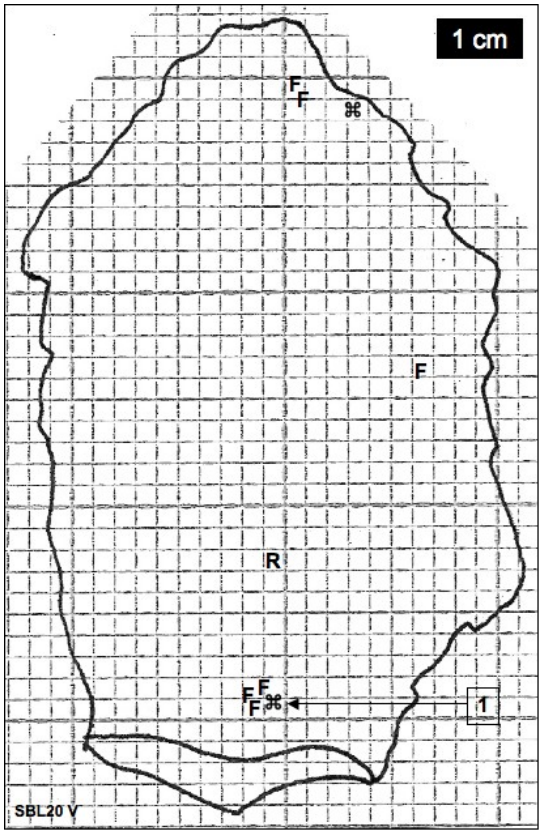


Figure D.96: Distribution map for SBL20, ventral side. For key see Figure D.1.

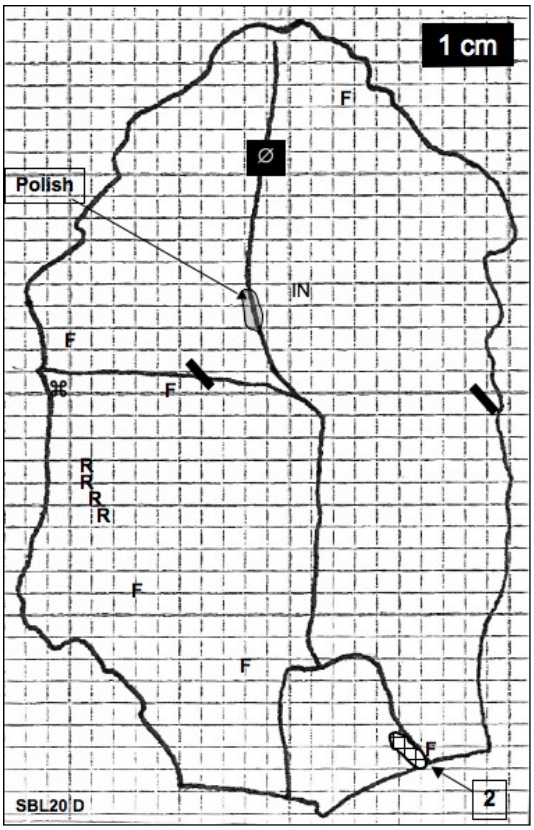


Figure D.97: Distribution map for SBL20, dorsal side. For key see Figure D.1.

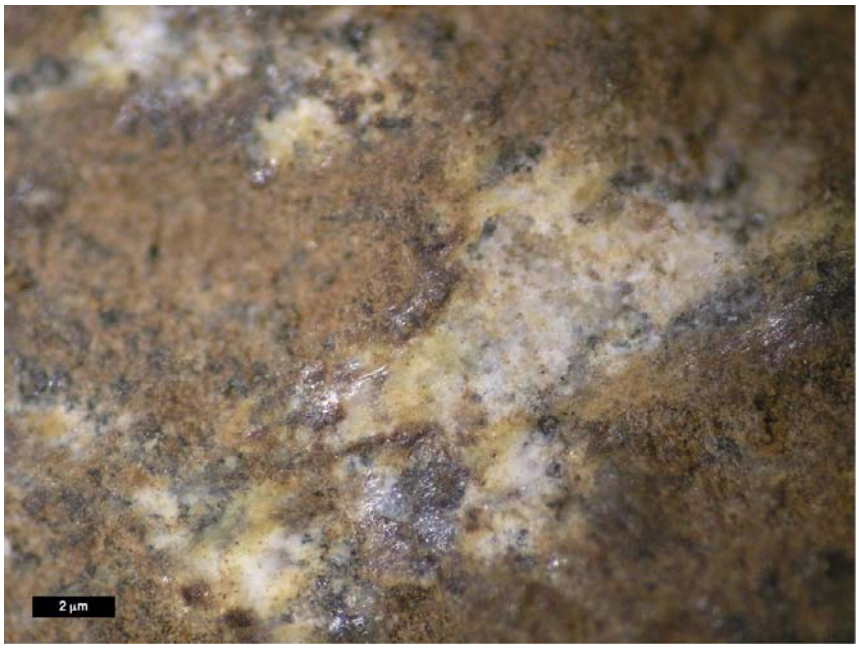


Figure D.98: 1= Micrograph of fat on SBL20. Magnification 50.

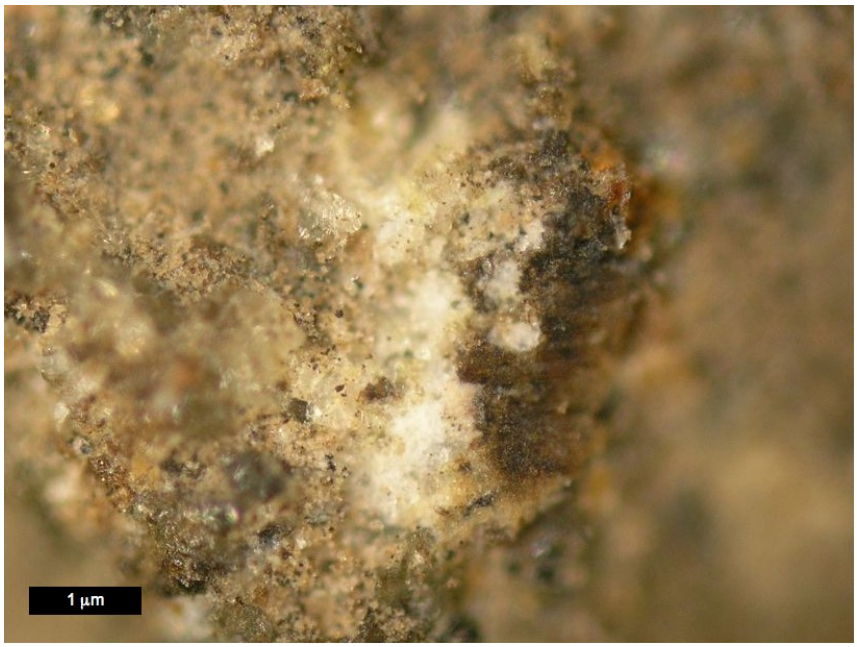


Figure D.99: 2= Micrograph of a "brown and white deposit" on SBL20. Magnification 100.

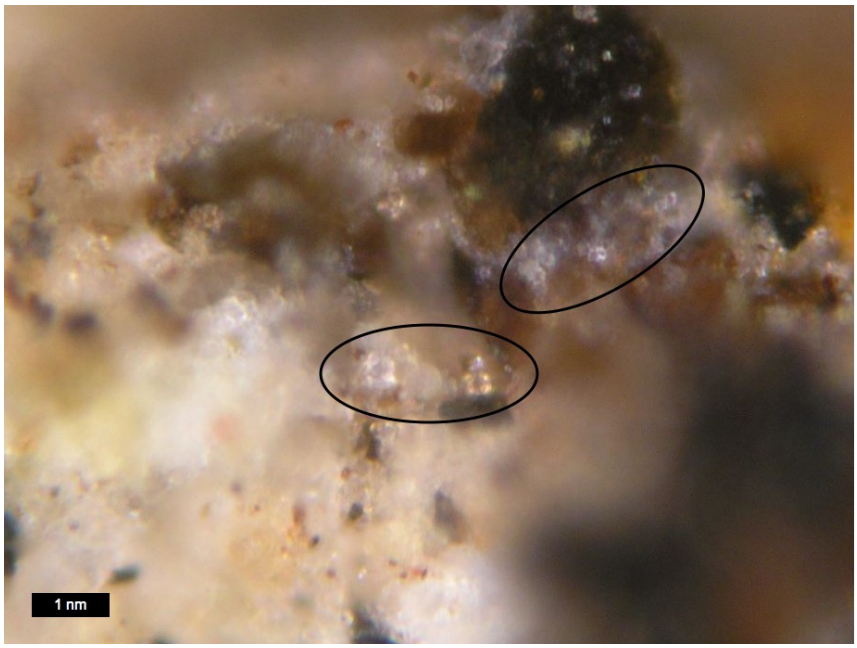


Figure D.100: 2= Micrograph of starch grains in the "brown and white deposit" on SBL20. Inside the circles are some clear examples of starch grains. Note the characteristic extinction crosses. Magnification 800, brightfield polarised light.

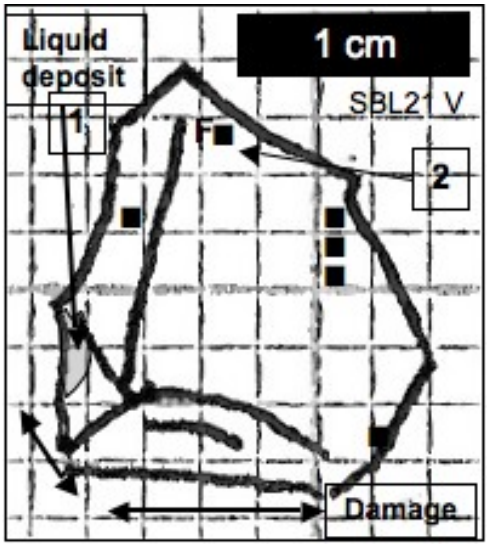


Figure D.101: Distribution map for SBL21, ventral side. For key see Figure D.1.

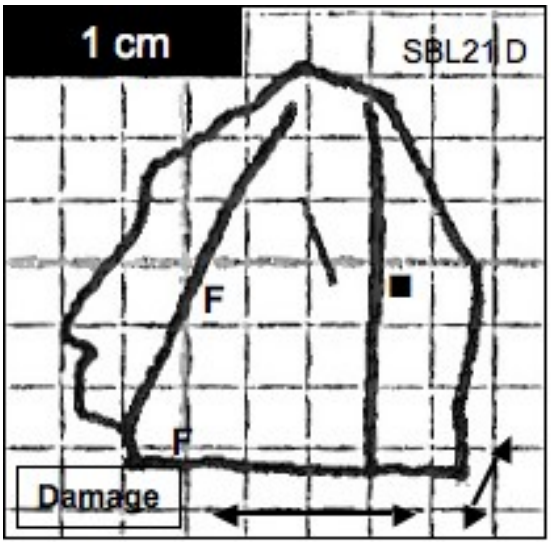


Figure D.102: Distribution map for SBL21, dorsal side. For key see Figure D.1.

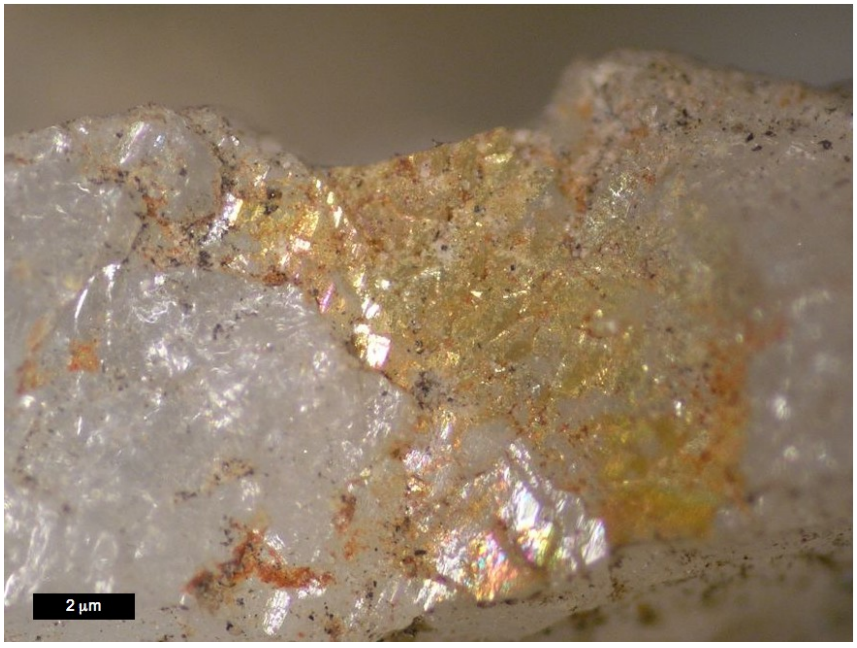


Figure D.103: 1= Micrograph of a dried liquid deposit on SBL21. The reflective parts of this yellow deposit appear to be an inclusion, but in Figure C.104 it is apparent that the deposit is a clear layer on top of the stone. Magnification 50.

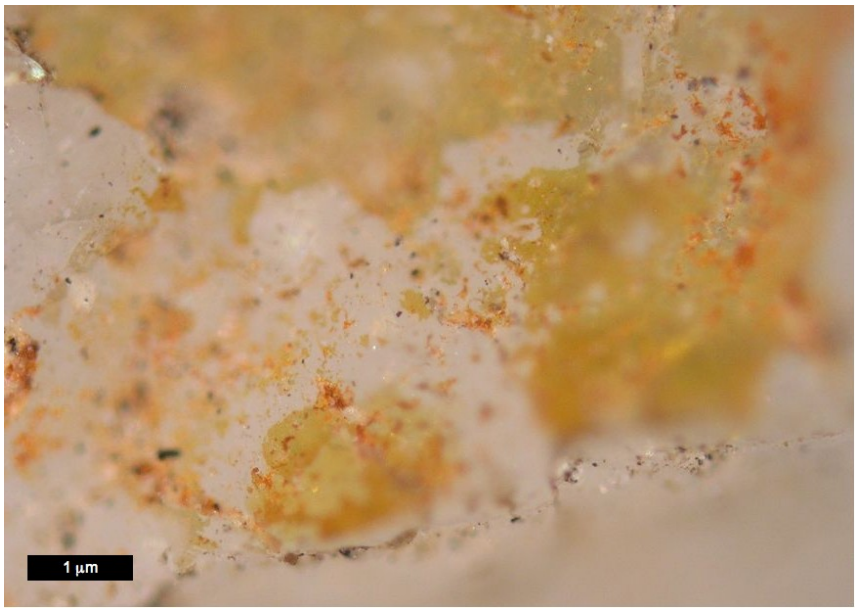


Figure D.104: 1= Micrograph of a dried liquid deposit on SBL21. Magnification 100, brightfield polarised light.

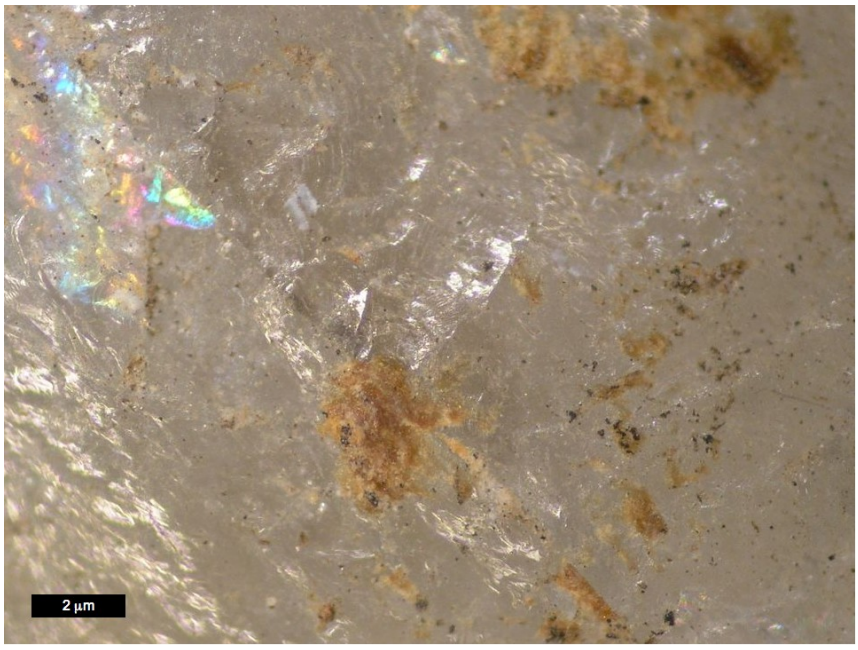


Figure D.105: 2= Micrograph of muscle tissue on SBL21. Magnification 50.

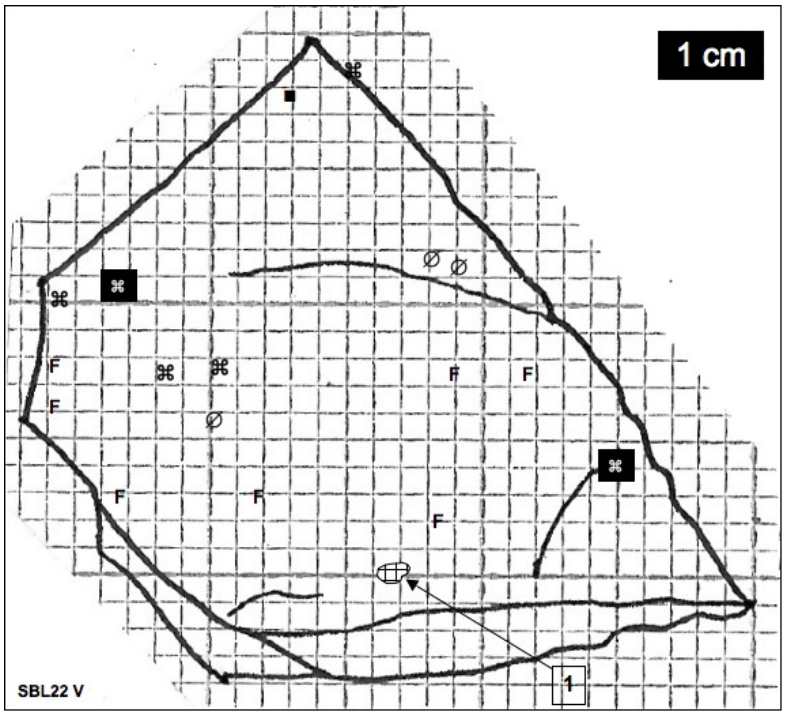


Figure D.106: Distribution map for SBL22, ventral side. The animal type residues are present in a suite, indicating that they could be use-related. For key see Figure D.1.

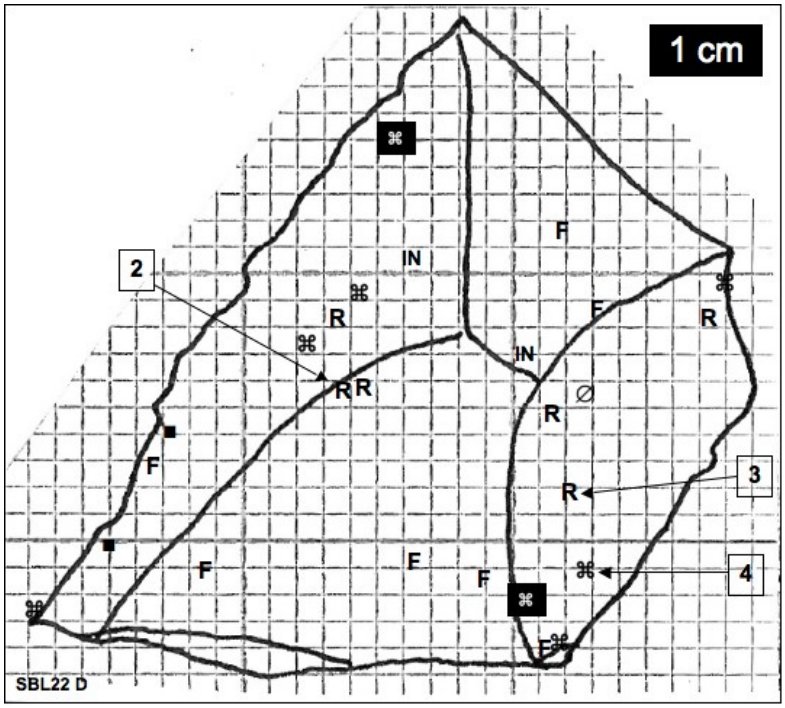


Figure D.107: Distribution map for SBL22, dorsal side. The animal type residues are present in a suite, indicating that they could be use-related. For key see Figure D.1.



Figure D.108: 1= Micrograph of a "brown and white deposit" on SBL22. Magnification 50.

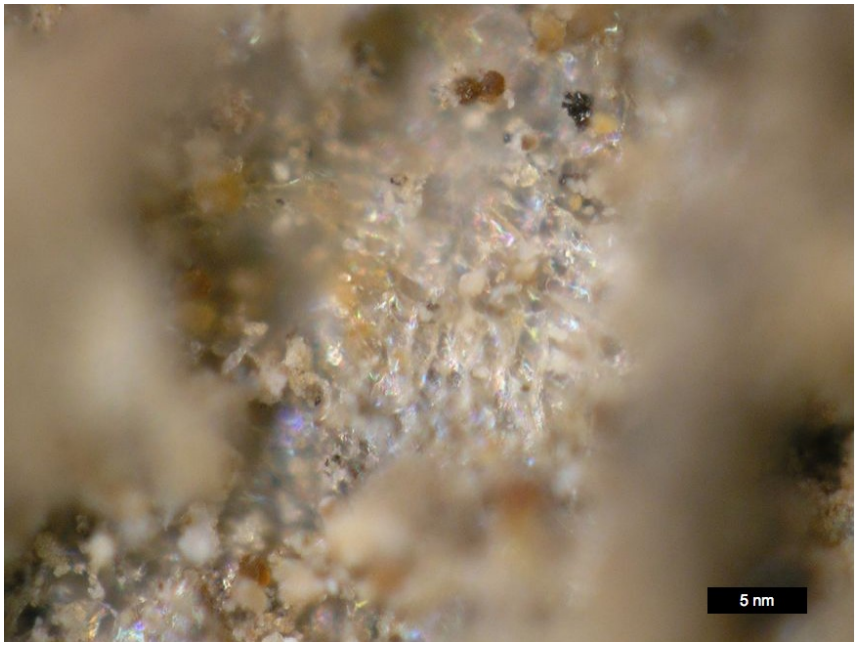


Figure D.109: 2= Micrograph of part of a rootlet on SBL22. Note the clear cells with cell walls. Magnification 200.

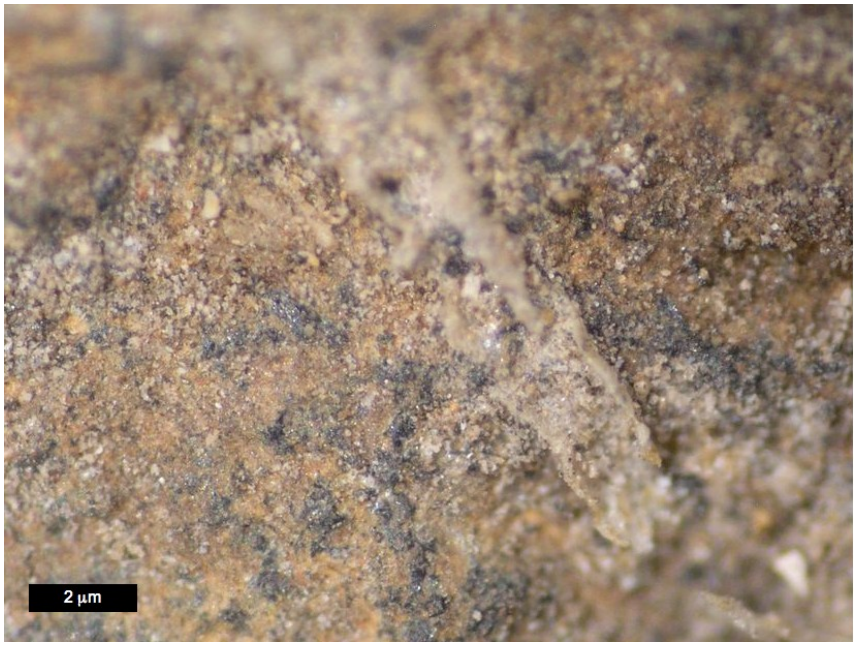


Figure D.110: 3= Micrograph of a rootlet on SBL22. Magnification 50.

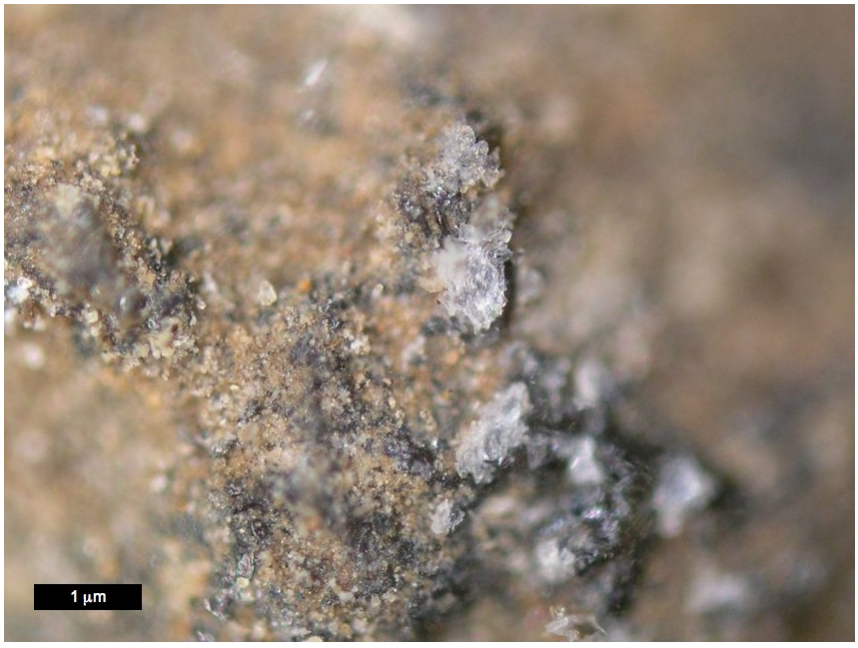


Figure D.111: 4= Micrograph of white bone on SBL22. Magnification 100.

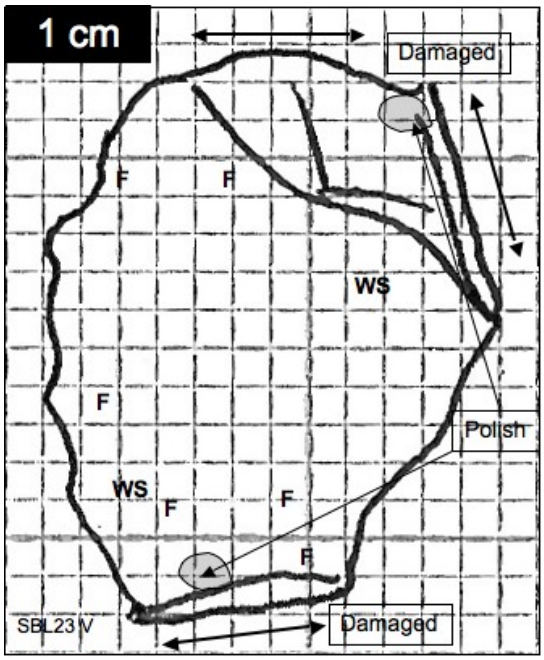


Figure D.112: Distribution map for SBL23, ventral side. For key see Figure D.1.

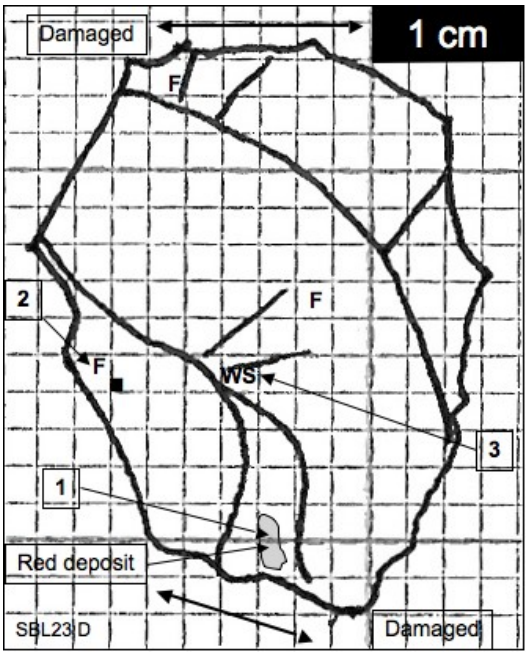


Figure D.113: Distribution map for SBL23, dorsal side. For key see Figure D.1.

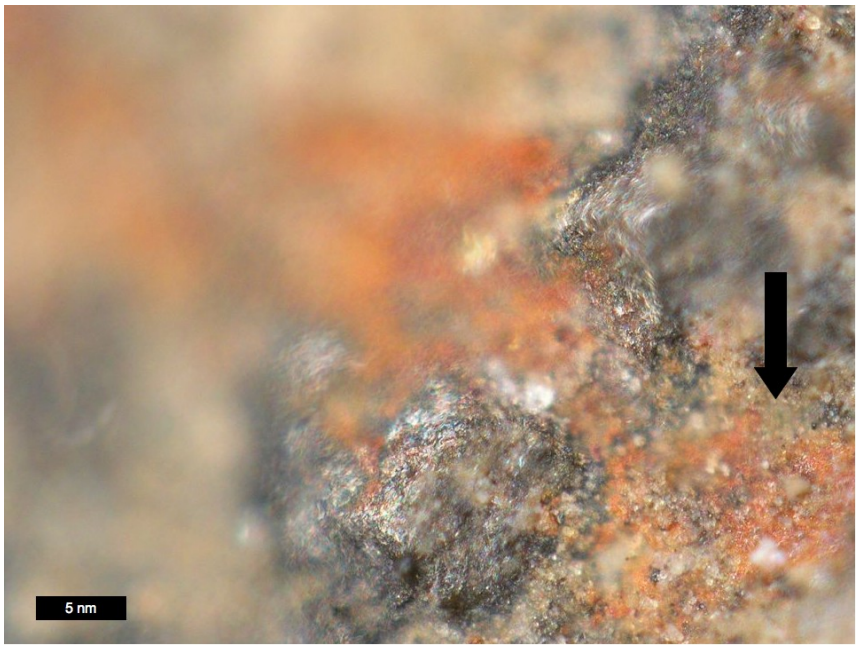


Figure D.114: 1= Micrograph of a red deposit on top sediment (indicated by the arrow). SBL23. Magnification 200.



Figure D.115: 2= Micrograph of fat on SBL23. The fat smear, which has bluish edges, is located on top of a sediment patch. Magnification 100.

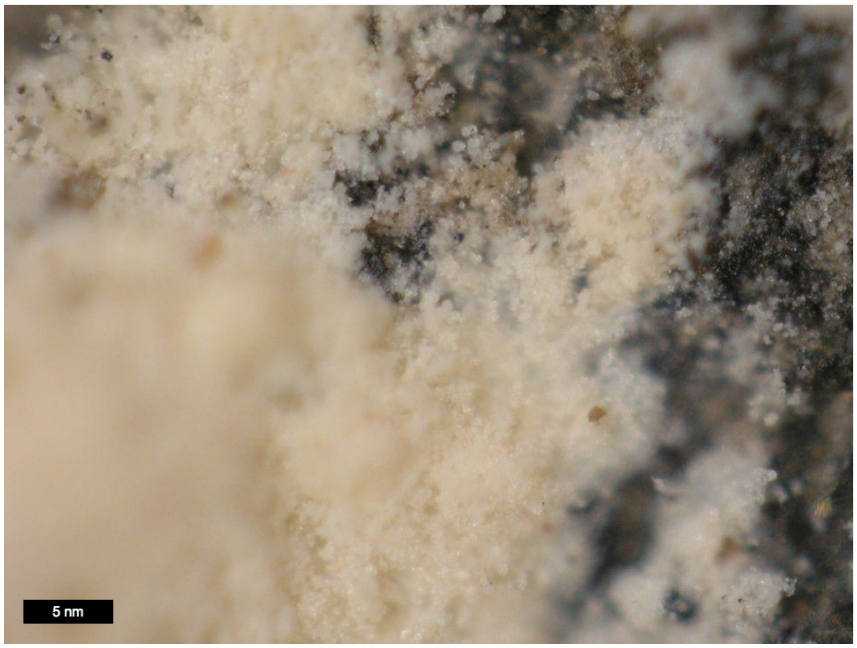


Figure D.116: 3= Micrograph of "white starchy" on SBL23. Note that granular nature of the deposit. Magnification 200.

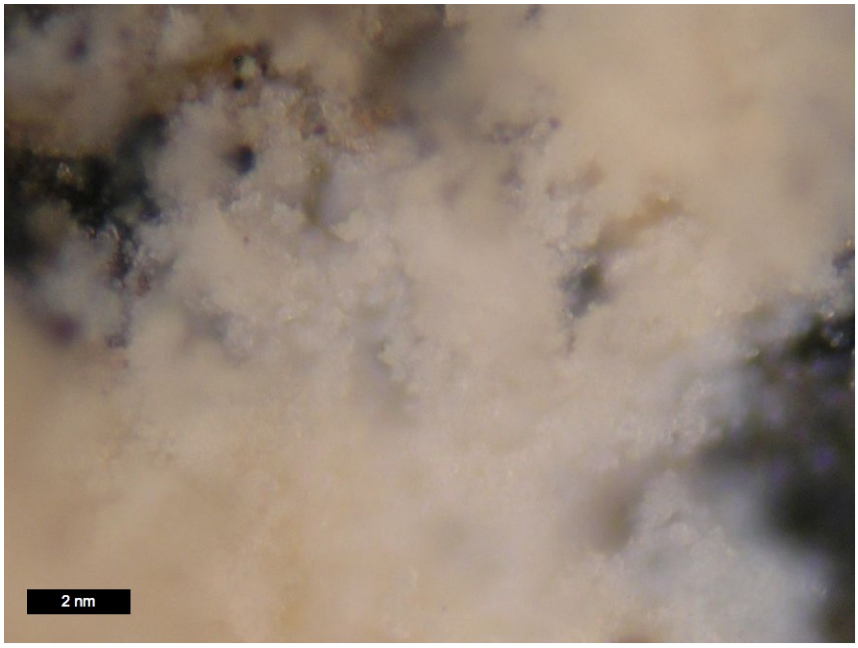


Figure D.117: 3= Micrograph of "white starchy" on SBL23. Note that granular nature of the deposit. Magnification 500, brightfield cross-polarised light.

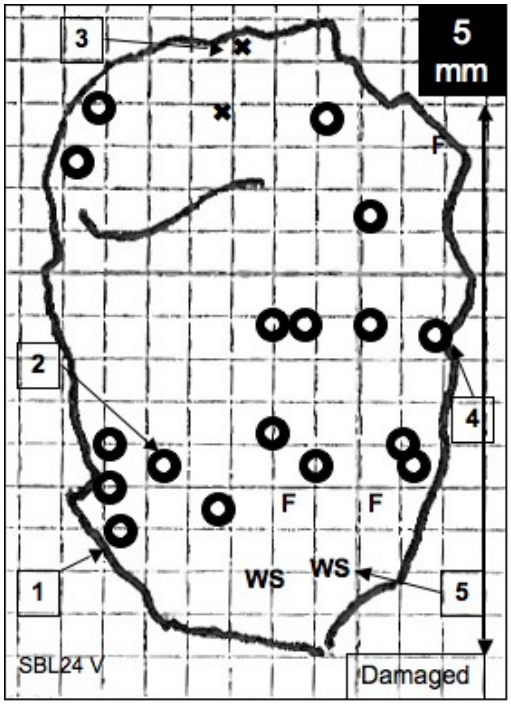


Figure D.118: Distribution map for SBL24, ventral side. For key see Figure D.1.

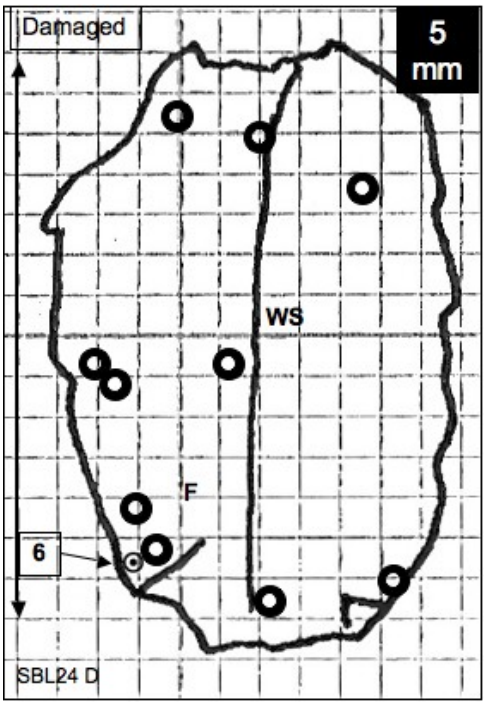


Figure D.119: Distribution map for SBL24, dorsal side. For key see Figure D.1.



Figure D.120: 1= Micrograph of an unidentified circular plant deposit on SBL24. Magnification 50.



Figure D.121: 2= Micrograph of an unidentified circular plant deposit on SBL24. Magnification 50.



Figure D.122: 3= Micrograph of a red fat-like deposit on SBL24. Magnification 100.

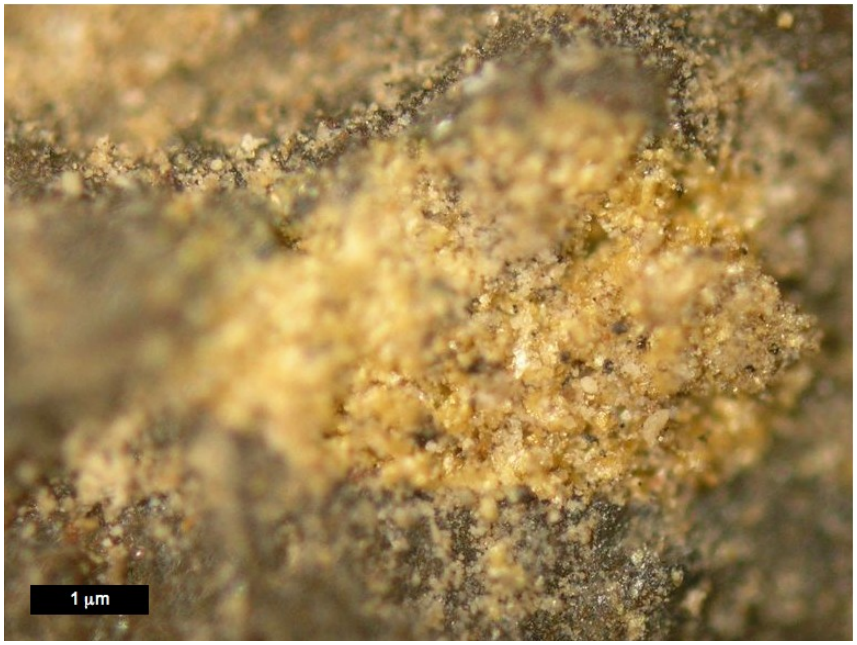


Figure D.123: 4= Micrograph of a circular plant deposit on SBL24. Magnification 100.

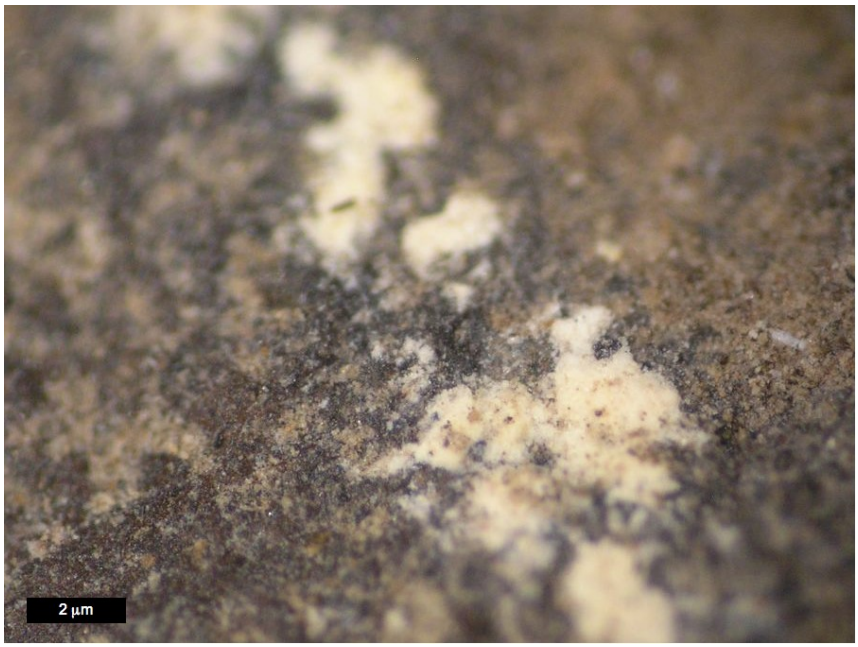


Figure D.124: 5= Micrograph of a "white starchy" deposit on SBL24. Magnification 50.



Figure D.125: 6= Micrograph of an unidentified circular plant deposit on SBL24. Magnification 50.

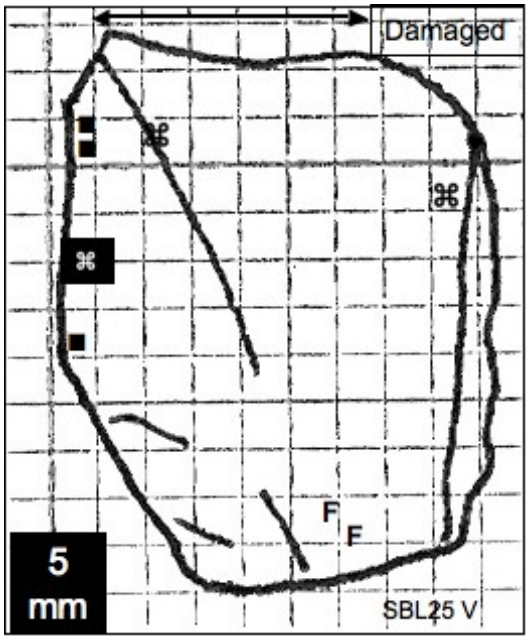


Figure D.126: Distribution map for SBL25, ventral side. For key see Figure D.1.

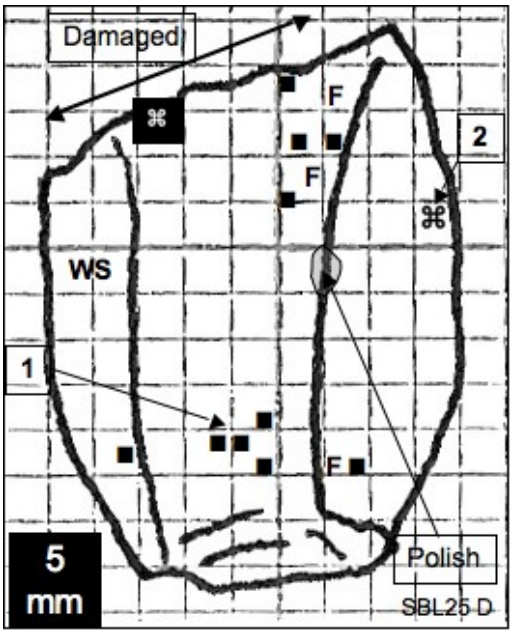


Figure D.127: Distribution map for SBL25, dorsal side. For key see Figure D.1.

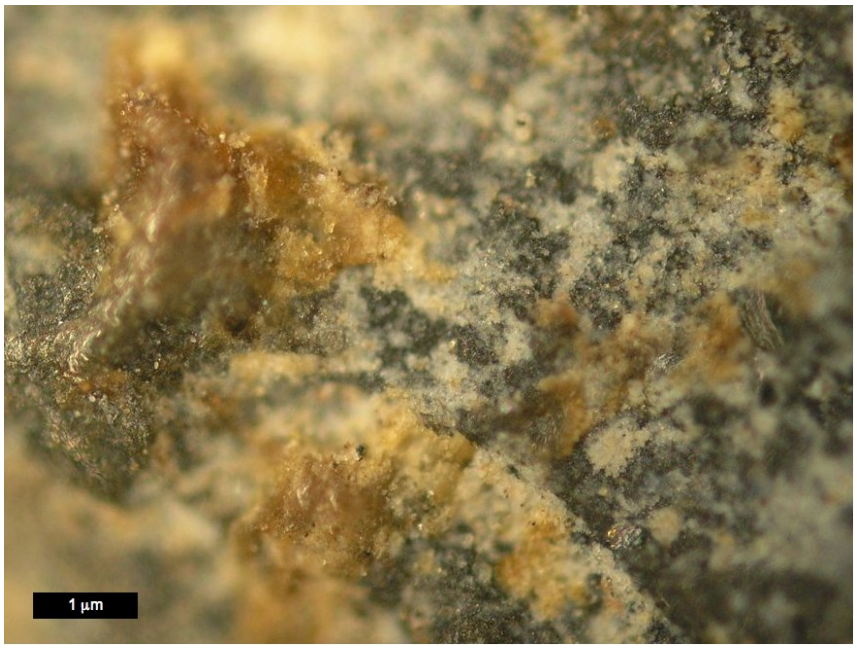


Figure D.128: 1= Micrograph of muscle tissue on SBL25. Magnification 100.

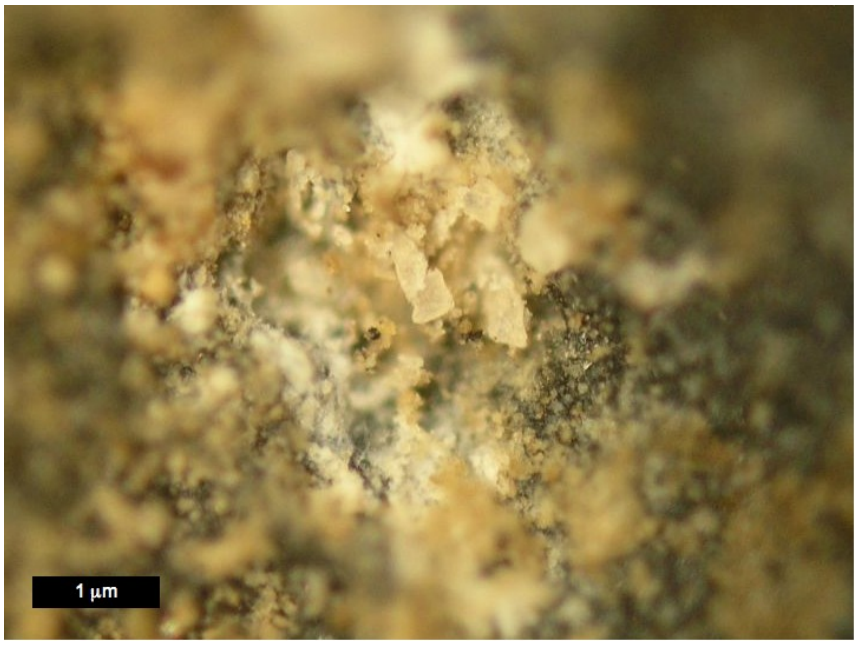


Figure D.129: 2= Micrograph of bone on top of sediment on SBL25. Magnification 100.

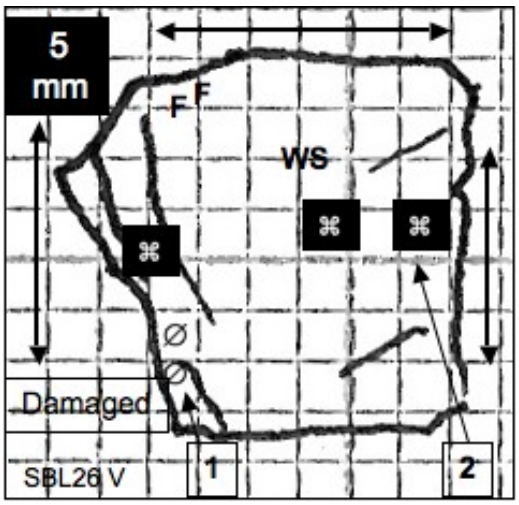


Figure D.130: Distribution map for SBL26, ventral side. For key see Figure D.1.

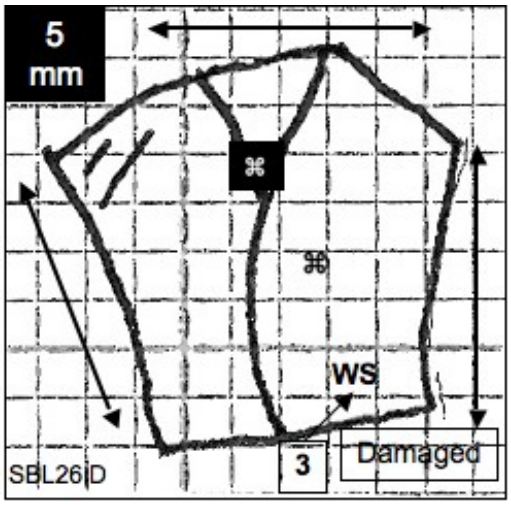


Figure D.131: Distribution map for SBL26, dorsal side. For key see Figure D.1.



Figure D.132: 1= Micrograph of plant tissue on SBL26. Magnification 50.

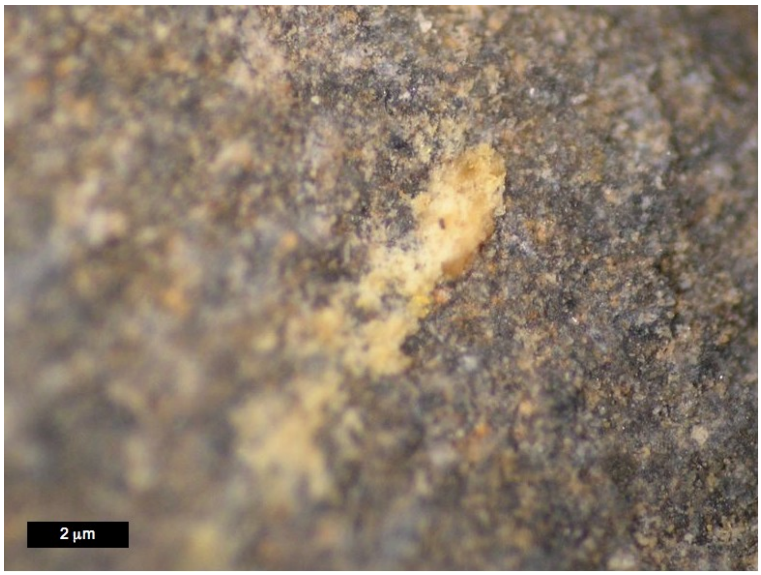


Figure D.133: 2= Micrograph of burnt collagen on SBL26. Magnification 50.



Figure D.134: 3= Micrograph of a "white starchy" deposit on SBL126 Magnification 50.

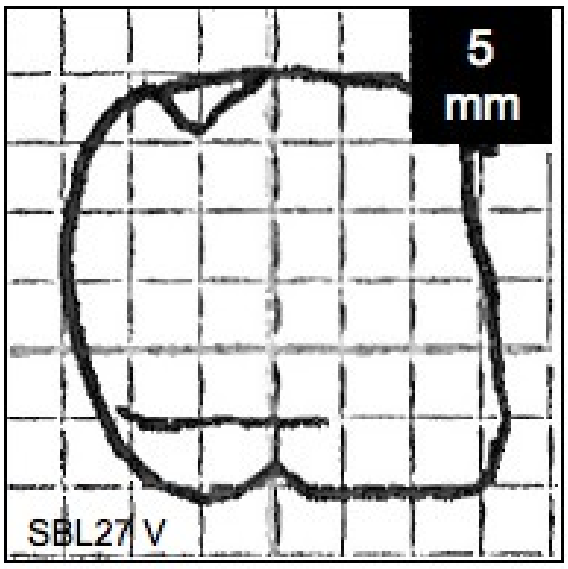


Figure D.135: Distribution map for SBL27, ventral side. For key see Figure D.1.

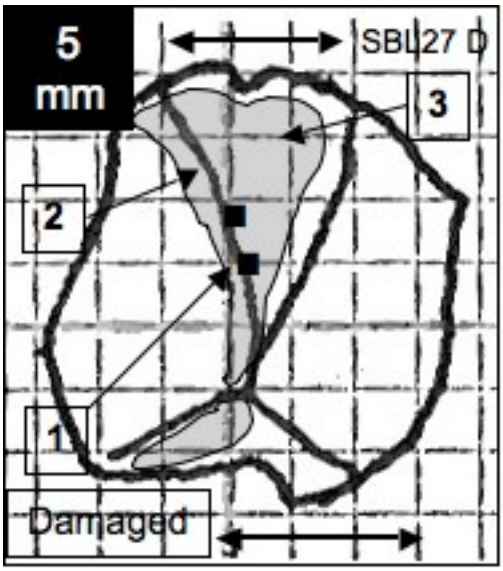


Figure D.136: Distribution map for SBL27, dorsal side. For key see Figure D.1.

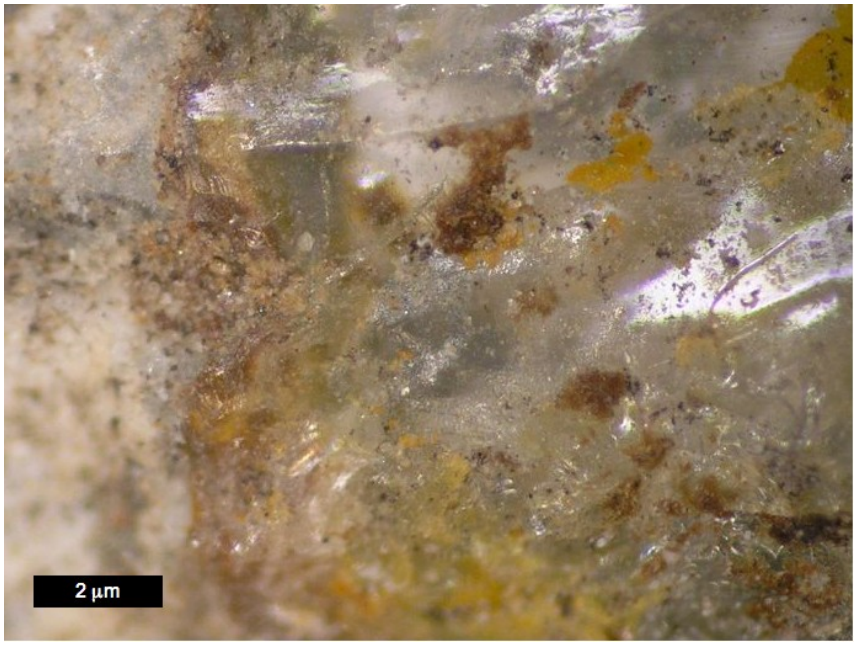


Figure D.137: 1= Micrograph of brown muscle tissue on SBL27. Magnification 50.



Figure D.138: 2= Micrograph of the yellow inclusion on SBL27. Note this picture is similar to the liquid deposit in Figures C.103 and C.104. However, Figure C.139 demonstrates that this is an inclusion and not a thin film on top of the stone surface. Magnification 50.



Figure D.139: 3= Micrograph of the yellow inclusion on SBL27. Magnification 200.

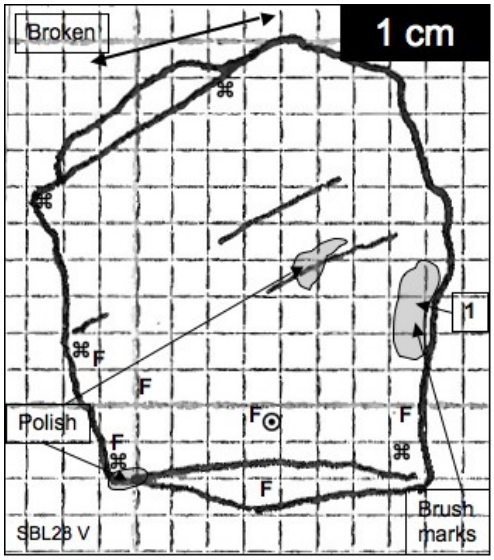


Figure D.140: Distribution map for SBL28, ventral side. For key see Figure D.1.

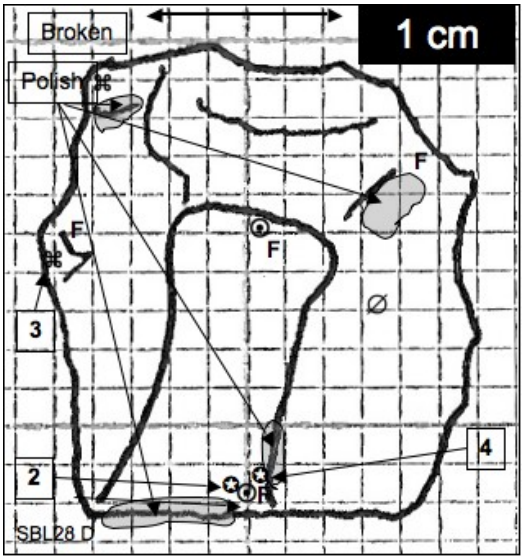


Figure D.141: Distribution map for SBL28, dorsal side. For key see Figure D.1.

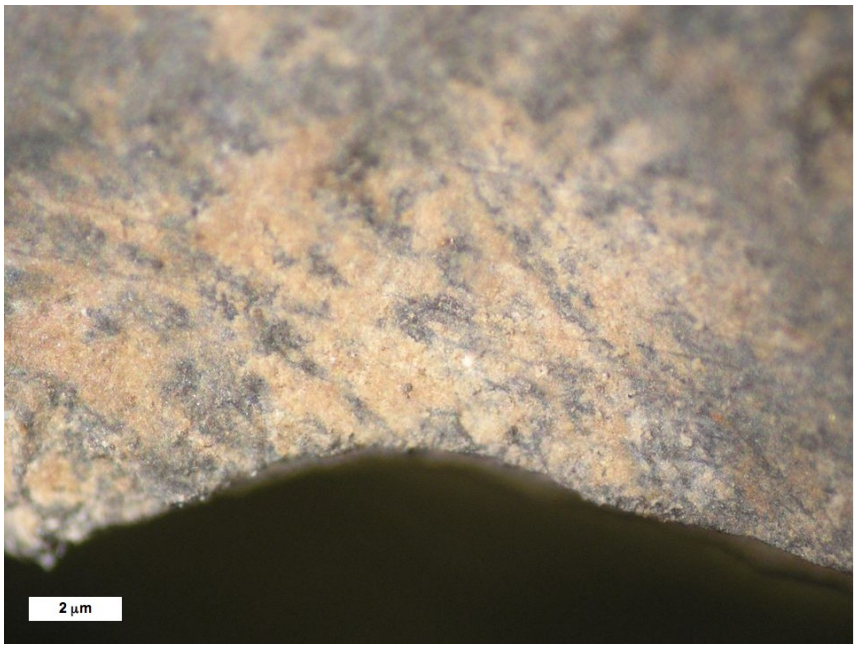


Figure D.142: 1= Micrograph of brush marks on SBL28. Note that the brush marks have a random orientation. Sometimes they can appear more ordered, in which case they can be mistaken for use-marks. Magnification 50.



Figure D.143: 2= Micrograph of an ochre mixture on SBL28. The white fibre could be a connective fibre, but this residue needs further investigation. Magnification 50.

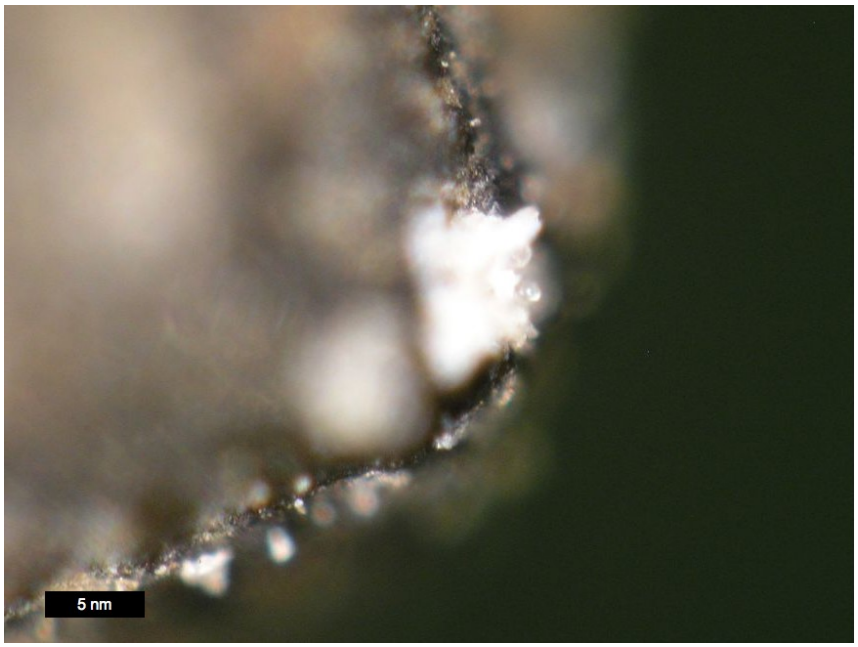


Figure D.144: 3= Micrograph of reflective bone on SBL28. Magnification 200.

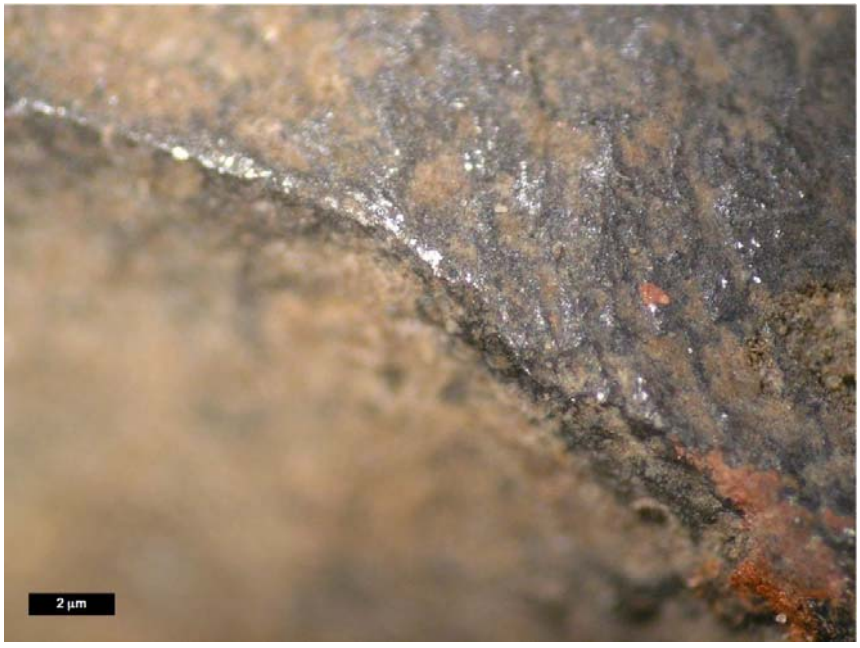


Figure D.145: 4= Micrograph of polish and an ochre mixture on SBL28. Magnification 50.

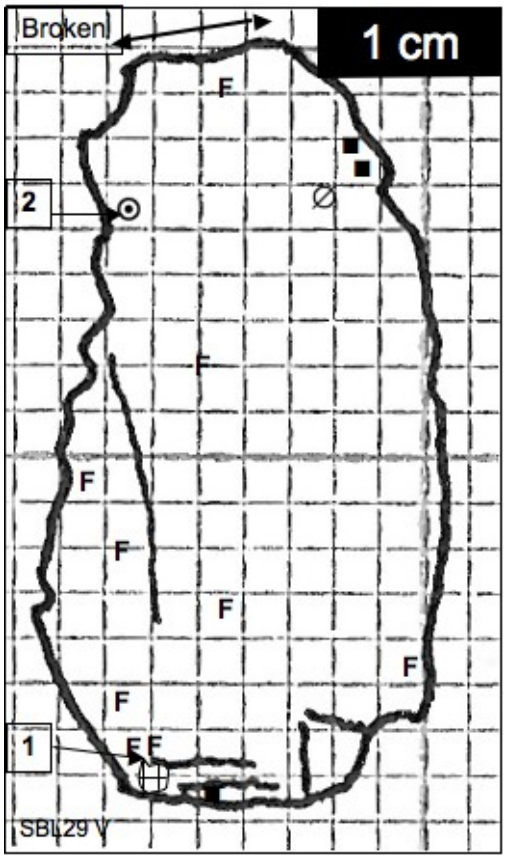


Figure D.146: Distribution map for SBL29, ventral side. For key see Figure D.1.

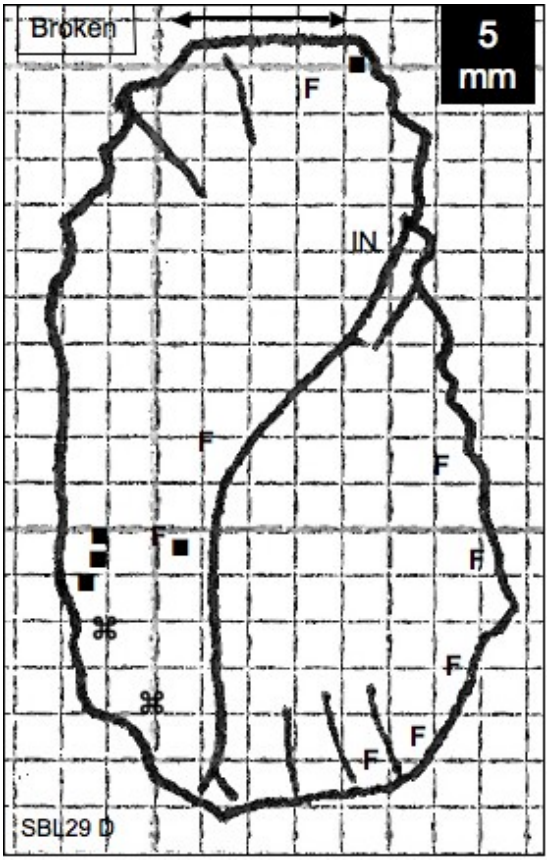


Figure D.147: Distribution map for SBL29, dorsal side. For key see Figure D.1.

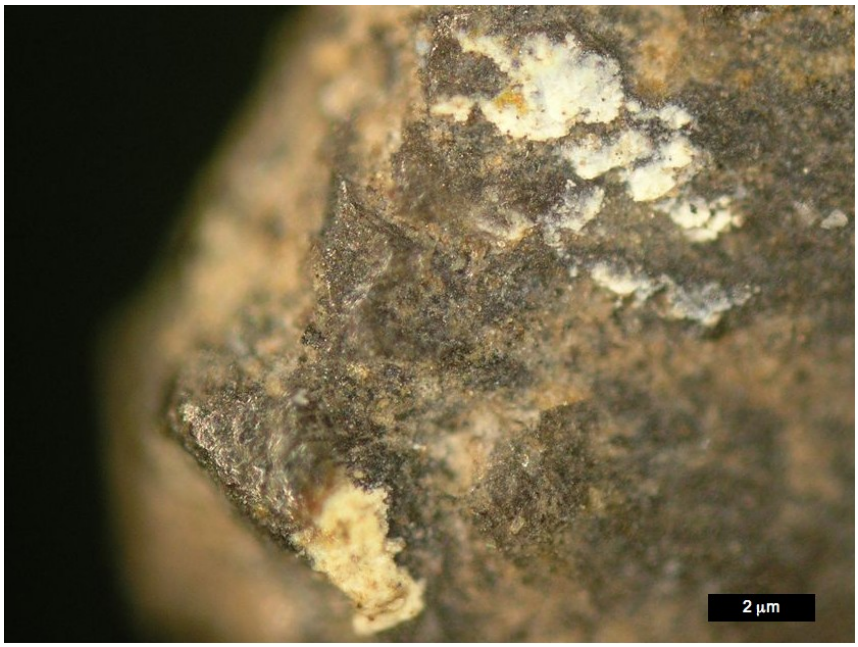


Figure D.148: 1= Micrograph of a "brown and white deposit" (bottom) and white/bluish fat (top) on SBL29. Magnification 50.



Figure D.149: 2= Micrograph of glassy resin on SBL29. Magnification 200.

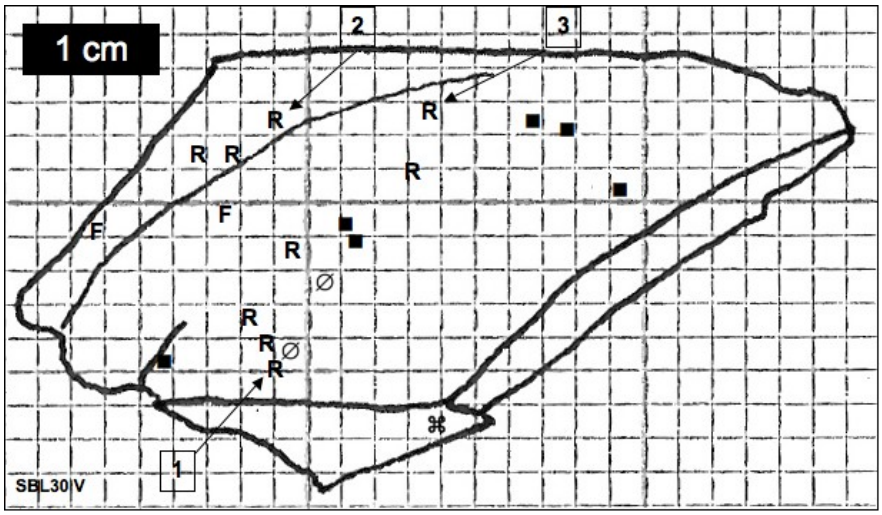


Figure D.150: Distribution map for SBL30, ventral side. For key see Figure D.1.

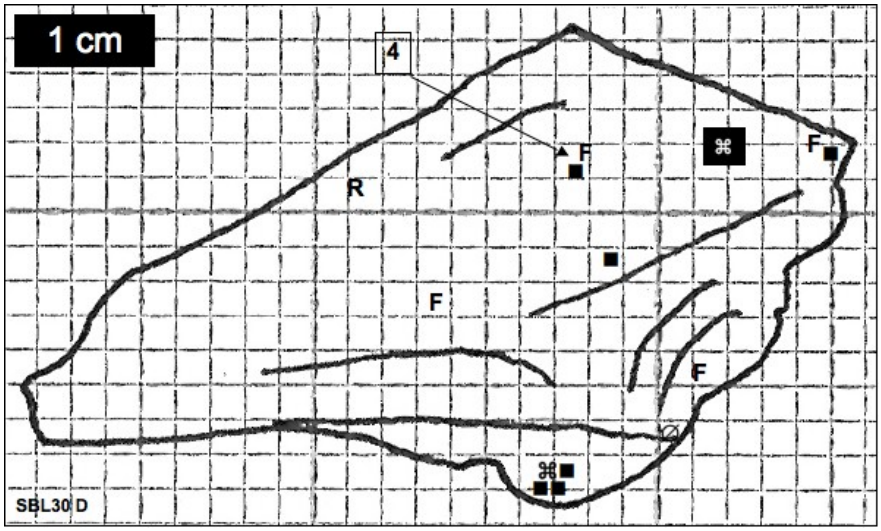


Figure D.151: Distribution map for SBL30, dorsal side. For key see Figure D.1.

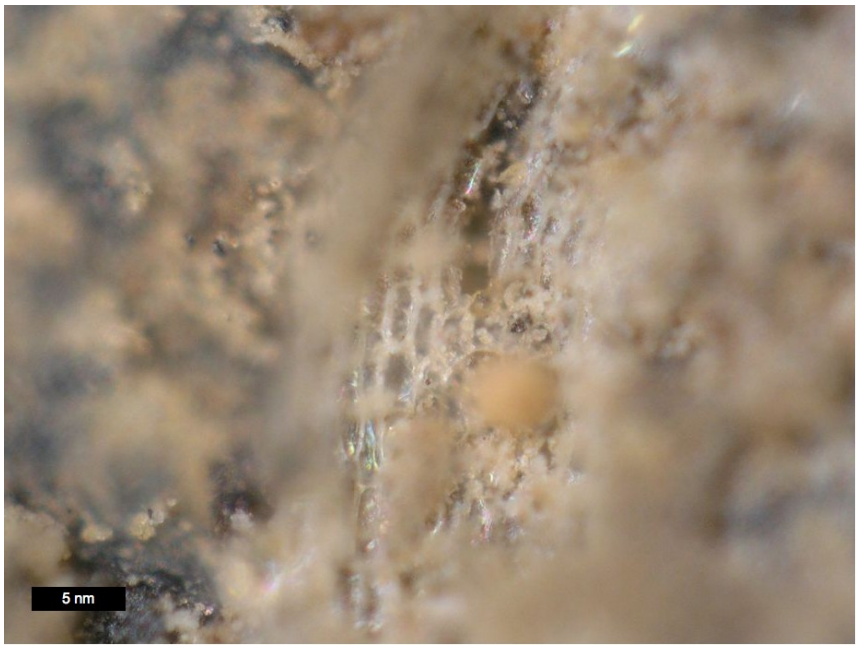


Figure D.152: 1= Micrograph of part of a rootlet on SBL30. Note the brick-like cell structure. Magnification 200.



Figure D.153: 2= Micrograph of a rootlet on SBL30. Magnification 50.



Figure D.154: 3= Micrograph of a rootlet on SBL30. Magnification 50.

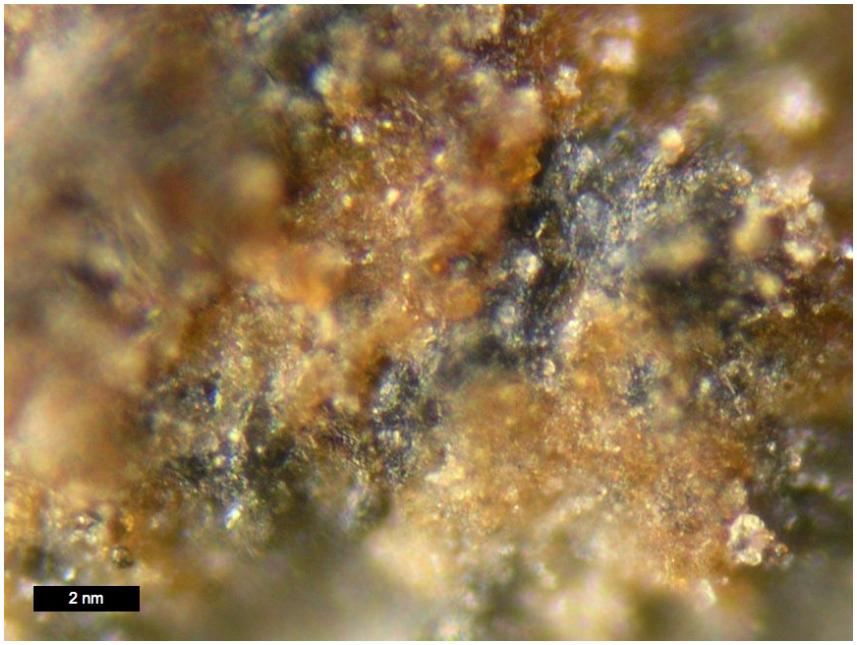


Figure D.155: 4= Micrograph of muscle tissue on SBL30. Magnification 500.