Care and Conservation of Natural History Collections

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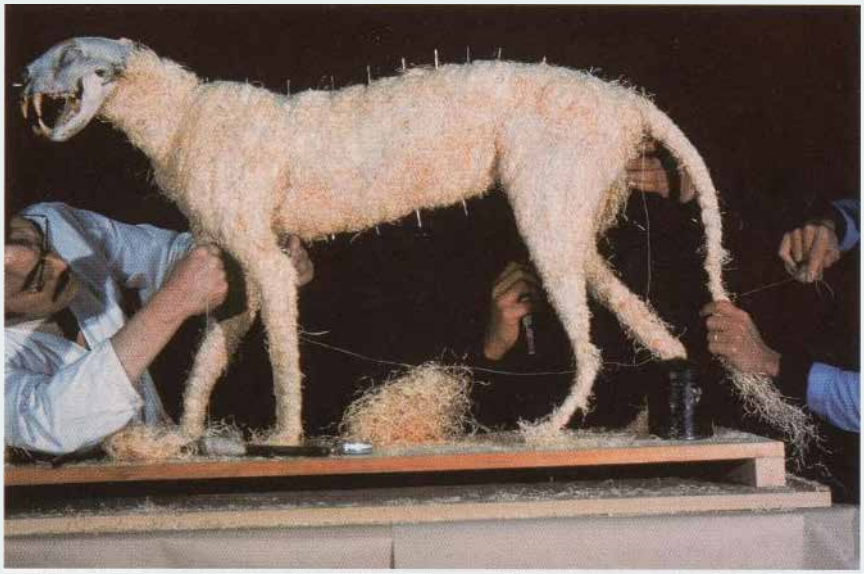

Plate 3 A traditional mannikin bound with wood wool. This technique was used to mount most large mammals in the UK prior to 1980 (Dick Hendry).

Plate 4 A mould being removed from a mounted hog, leaving the real hair embedded in a fibreglass body (Dick Hendry).

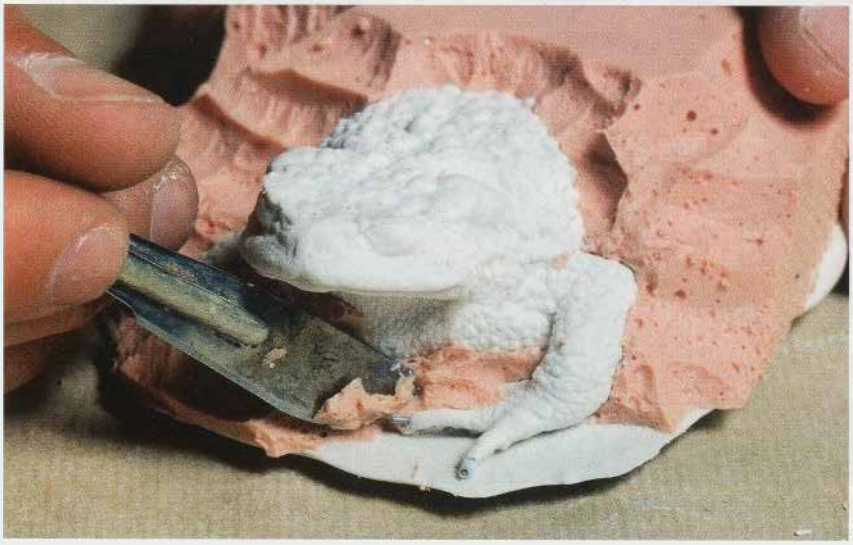

Plate 5 A plaster cast of a toad being removed from a dental alginate mould (Dick Hendry).

1

Vertebrates

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Introduction

Historically, there has been little serious scientific research into methods for preparing, preserving and conserving dry vertebrate collections. Techniques progressed because they were faster, cheaper or resulted in aesthetically pleasing collections. The longterm conservation of the specimen was often a secondary consideration. There are many reasons for this, all associated with the perceived value of natural science collections. In the past ten years, however, considerable efforts have been made to explain the value of such collections culminating in Duckworth *et al.* (1993), which opened up the debate and suggested proposals for the way forward.

The awakening of interest in the care of natural science collections, led by the Society for the Preservation of Natural History Collections (SPNHC) in North America, has resulted in a shift in the allocation of resources. Much more emphasis is now being placed on preventive conservation as a cost-effective way of providing collection care.

A preventive conservation strategy, however, should be supported by a parallel research programme investigating methodologies. A knowledge of how dry vertebrate collections were prepared in the past is essential to determine the efficacy of previous methods, and to focus limited resources on the best techniques for safe handling and longterm storage today.

Specimen preparation and handling

There are important health and safety issues relating to the handling of dead vertebrate material. Richards (199-i) describes both the legislation and hygiene precautions in this area, and suggests a sensible code of practice for natural history departments. It is important that all staff likely to come into contact with such material are made aware of the need to maintain a safe working environment.

Primary specimen care

Receiving specimens

Museums usually receive whole vertebrate specimens either freshly dead or in a deep-frozen condition. Such material should be examined and dealt with in the laboratory area where the recommended hygiene precautions may be observed.

External parasites are more easily observed on fresh specimens, and may be transferred, on a fine artists' brush, into a container of 70% industrial methylated spirit (IMS). Dipping the brush tip into chloroform will help to pacify lively parasites, but this procedure must be carried out within a fume cupboard. Alternatively, with birds and mammals, the fur or feathers may be brushed on to a sheet of plain paper and the specimens collected using a pooter aspirator.

Although Florian (1990) argues that freezing natural history specimens may reduce their research potential, for most institutions this is an inevitable part of acquiring specimens in reasonable condition. If donated material is likely to deteriorate, the best option is to wrap it in polythene and deep-freeze it before transit. The freezer compartment of a domestic refrigerator will suffice for small specimens, whilst a domestic deep-freezer will keep wrapped specimens in good condition for many months. Before dispatch it is preferable to remove the polythene, wrap the specimens in sufficient absorbent paper to contain any fluid leakage during transit, and send in a stout cardboard or polystyrene container. Specimens sent this way are less likely to deteriorate than those sent in polythene bags. If, however, the specimen has started to deteriorate, is likely to be delayed in transit, or is not wanted strictly for taxidermy purposes, then it would be wise to seal it in a polythene bag. Any documentation, except a water-resistant, tie-on label, should be sent in a polythene wallet attached to the outside of the container. Donors should be advised on when to send frozen material to ensure that staff will be available to receive it.

The receiving institution should have procedures in place to deal with frozen material efficiently. A well labelled area in a large freezer or separate `recent acquisitions' freezer will help to ensure that specimens do not get lost, and that the initial documentation procedures, including taking weight and standard measurements, are carried out.

The condition of the specimen when it arrives in the museum may determine its fate. If decomposition is at an advanced. stage, it may still be possible to salvage osteological material and samples for DNA analysis, a growing area for the destructive use of museum specimens. Such samples are best taken from areas of the body, such as subcutaneous muscle tissue, which are less likely to have been in contact with external contaminants. Freeze-drying is the only option if it is imperative to dry-preserve the material. Specimens required to be prepared by traditional taxidermy techniques, as either mounted specimens for exhibition or study skins for the reference collections, must be reasonably fresh. Assessing the true condition of specimens which visually appear to be suitable for mounts or study skins comes with experience, but there are simple indicators of decay. The skin around the eyes of birds and mammals is the first place to look. Gently stroking this area will tell you if the specimen has begun to deteriorate. If the fur or feathers begin to come away (slipping), a little powdered alum rubbed into the skin may prevent further loss. Any such damage, especially in the head area, will make the specimen less suitable as an exhibition mount. Study skins, however, may be prepared from all specimens which survive the skinning and cleaning processes relatively intact.

Specimens, on arrival, may be soiled with blood, usually from the bill, nose or throat. These areas should be plugged with cotton wool to prevent further soiling and the blood, if fresh, removed at this stage. There is no consensus as to the best method. Hangay and Dingley (1985) recommend swabbing with cold water and adding a little ammonia for stubborn stains, whilst Sutton (1916) and Clancey (1959) prefer using warm and hot water respectively. Chemicals such as hydrogen peroxide or, as Dill (1957) suggests, oxalic acid should only be used if it is imperative to remove the blood – and then try dilute solutions first (see also Cleaning bird skins, p. 5).

Specifications for a large freezer and storage

If specifying for a walk-in freezer to hold all types of vertebrate material, ensure that the internal temperature is at least -20°C. The door handle must be fitted with an internal emergency release mechanism, and both visual and audible high temperature alarms should be installed, the latter linked to a telephone or security control. The telephone number of the 24-hour emergency maintenance firm should be clearly labelled on the freezer door. A large freezer will maintain an adequate internal temperature for at least 12 hours following a breakdown. It is good practice, however, to make a separate arrangement with an outside commercial freezer firm in the event of an emergency. This should be part of any disaster plan.

Shelving units in the freezer, such as those recommended by McConachie (1992), help

organize the space efficiently. For safety reasons, large frozen specimens should not be stored on shelves above head height. They can be stored safely in strong polythene bags suspended from butchers' hooks in the walls and ceiling. Colour-coded polythene bags and labels in industrial-weight plastic wallets reduce the risk of specimens being discarded with the refuse.

Skin preservatives

Williams and Hawks (1987) have listed the variety of chemicals used in the preparation of mammals, alongside a list of their users. Their aims were to identify hazardous chemicals and assess the suitability of certain collections for future research. Such lists are useful starting points before embarking on the more sophisticated spot tests and microanalysis that Found and Helwig (1995) have investigated.

There appear to be no preservatives for vertebrate skins which prevent insect attack in the long-term, and preventive measures are a better solution. Over the past 200 years, however, many different preservatives and combinations have been tried, a few achieving world-wide usage.

Arsenic and mercury

Although, as Giitebier (1989) reports, arsenic (dry, white arsenic trioxide) was advocated for use on specimens to deter attack from insects as long ago as the seventeenth century, it was its rediscovery as arsenical soap in the middle of the eighteenth century that led to its universal use for the next 200 years. Farber (1987) suggests that this discovery was a key element in the development of ornithology.

In the early days its strongest rival, and sometimes companion, was mercuric chloride (corrosive sublimate). Browne (1886) and Davis (1907) describe the common practice of applying arsenical soap to the flesh side of the skin and spraying the fur or feathers with the mercury-based compound. Such were its advocates that Rowley (1925) warned collectors not to offer specimens to museums unless they were preserved with arsenic.

The dangers of using such toxic chemicals were well known and highlighted by many preparators including Ward (1906), Hasluck (1914) and, most vociferously, Pray (1943). However, up to the mid 1960s writers such as Anderson (1965) still reflected the earlier perception that arsenic was merely a nuisance rather than a considerable hazard to health.

There is no doubt that these chemicals did work, at least in the short term. To suggest as Howie (1985) does, that their use is the reason for the shortage of specimens over 100 years old, is to misunderstand the problem. The most important part of dry vertebrate specimen care is not the preservative chemical but the aftercare. Batty (1885) recognized the prime importance of keeping pests away from specimens by enclosing them in well sealed containers which he called `tight chests' — an early advocate of preventive conservation.

Although mercury fell out of favour in the twentieth century (the *British Musetun Handbook for Collectors,* anon., 1904, suggested that it made bird skins brittle), arsenic was popular and advocated in most standard textbooks, such as Mahoney (1973) and Moyer (1979), until the late 1970s. It still has pockets of support today and, in a series of tests on mammal skin preservatives by Hanacziwskyj *et al.* (1991), it was found to be the only one which did not damage deer skin collagen. Morris (1982) showed that the average lifespan of taxidermists using arsenic was not dramatically curtailed. Reports of its possible carcinogenic risks, such as Haneke (1977), may see its demise as a specimen preservative.

A useful spot test for the presence of arsenic on specimens can be found in Hawks and Williams (1986b).

Borax and alum

The most popular alternative to arsenic and mercury compounds was borax (sodium tetraborate), strongly promoted as a moth deterrent by Pray (1943). Used on its own, or mixed with a variety of other chemicals, it became the standard museum `safe' preservative, recommended in popular texts such as British Museum handbooks (anon., 1968) and Wagstaffe and Fidler (1968).

Whether it is an effective long-term moth deterrent is not clear. If used specifically as directed by Pray (1943) — who later recommended an improved formula with added formaldehyde, (Pray, 1951) — it gained the credibility of experienced taxidermists such as

Moyer (1981). Anderson (1965) even suggested that mixing it with arsenic rendered the latter `comparatively safe'. Whatever its insect-proofing qualities, it is still popular today as a drying agent and absorbent in bird skin preparation, although there is a growing group of preparatory who prefer not to use any preservation chemicals (see also Preparation of bird skins, p. 5).

Potash alum (potassium aluminium sulphate) was and still is used for preserving small to medium animal skins. Its astringent nature makes it very useful for halting small local areas of decay. Both Ward (1906) and Anderson (1965) maintain that this astringency makes it unsuitable for the relatively thin skins of birds, causing them to dry so hard that subsequent relaxation is very difficult.

Acids and tanning baths

Most large mammal skins used for exhibition mounts or in reference collections have either been cured with salt and preserved in acid pickling baths or subjected to a tanning process. Phenol (carbolic acid) was one of the most widely used pickling acids, either on its own or together with salt, alum or saltpetre.

Salt and sulphuric acid and salt and alum tanning were very popular methods for preserving mammal skins for museum collections. Today organic acids such as oxalic and formic have largely replaced sulphuric which, by remaining in the skin, may cause long-term deterioration. Formaldehyde together with Lancrolene oil was a well known basic tanning combination in the 1960s, and Mahoney (1973) recommended it, particularly for snake and lizard skins.

According to Hawks *et al.* (1984) techniques are available to determine how a skin has been tanned. Although these procedures are expensive, they may be essential in future to warn researchers of chemical preparation techniques which can degrade or destroy DNA ,and so avoid potentially unrewarding analysis.

Proofing chemicals to prevent attack by insects

Eulan U33 and Eulan BLS (Edolan), liquid moth- and beetle-proofing chemicals (Permethrin derivatives) used in the German wool and carpet industry, were first proposed by Rau (1968) for museum use. Although the results of a study by Arevard *et al.* (1981) on the effectiveness of these chemicals were inconclusive, for long-term preservation, more promising results were obtained by Funk and Sherfey (1975), Granqvist (1982) and Graf (1984).

Techniques for their use are discussed in Namlik (1975), Philips (1980), Philips and Philips (1981) and Septon (1987), and for several years Eulan derivatives were seen to be the solution for controlling insect damage to skins, mounted specimens and freeze-dried material. The USA has now phased out these chemicals, as they were suspected of causing damage to fish stocks when entering river systems. Eulan WBP, however, is still currently available in the UK.

The chemical which may replace Eulan is Mitan FF. Initial tests by Connelly and Rogers (1995) suggest that, when used at temperatures compatible with bird and mammal skins, it does offer freeze-dried mammals some protection against dermestid beetle attack. In the same experiment, freeze-dried birds proofed with Mitan FF fared no better than their unpreserved controls. The technique involves soaking skins in the chemical, at temperatures ranging from 100 to 140°C, for periods of up to an hour, followed by immersion in acetic acid solution. It is a very invasive technique and, because of the danger of bird and mammal skins deteriorating at these temperatures, probably only suitable for use with very fresh material.

Colour changes in fur and feathers due to preservatives and preservation techniques

Variations in the subtle colours of pelage and plumage have been used in taxonomic studies, particularly at the subspecies level. Several researchers, notably Howell (1937), Burns (1952) and Coetzee (1985), have suggested that preservative chemicals used during skin preparation may cause significant changes in the colour of plumage and pelage. Indeed, Hall (1937) emphasized the vital importance of documenting the preservatives used in museum collections because of this problem.

In a comprehensive study of chemicals used for mammal skin preparation, Burns (1952) found that a mixture of saltpetre and potash alum was the least likely to affect the colour

of fur. Chemicals not recommended were borax, mixtures of salt and alum, ethyl alcohol and carbon tetrachloride. Borax was found to be satisfactory for fresh skin preparation but unsuitable if the skin was later relaxed by water, a finding which coincides with the work of Downing (1945). Most workers in this field agree that colours such as red/browns are particularly susceptible to change, but the degree of change may vary with species.

Rogers and Daley (1988) point out that whilst chemicals, such as those in Huber's fluid (see Formulae at the end of this chapter, p. 36), have been criticized for over 150 years, they are still included in most of the standard texts. In a study on the preparation of bird collections, they concluded that all preservative chemicals tested caused some negative changes to either the colour or the form of at least one feather type. Salted bird skins suffered less colour change than those dried with borax, and were considered easier to relax. A petroleum spirit based thinner for paint and varnish sold in the USA as VM and P Naphtha (varnishmakers and painters' naphtha) was recommended as the most efficient, least toxic solvent, although it is suggested that the best practice is to substitute solvents with detergent solutions whenever practicable.

Preparation of bird skins

Informative accounts of collecting, taking scientific measurements, ageing and sexing are to be found in the standard scientific texts. Chapin (1940), Anderson (1965), Wagstaffe and Fidler (1968), Harrison (1976) and Hangay and Dingley (1985) are the most comprehensive. Skinning techniques are also illustrated in these texts and in taxidermy manuals such as Kish and Jonas (1976), Schmidt (1977) and Metcalfe (1981).

There are interesting variations in methods of preparing bird skins, some of which have a bearing on their long-term conservation.

Skinning

Traditionally, most birds are skinned through an incision along the breast and abdomen or under the wing. In Paulson (1989) it is recommended that downy young and adults with complex feather patterns should be skinned ventrally to avoid disturbing these patterns, whilst Halford (1987) suggests case-skinning birds (see p. 8) for similar reasons. Ducks and geese are usually skinned along the back to help reduce feather soiling when removing fat from the breast skin. Skinning techniques for non-traditional skin preparations are illustrated in Rogers and Wood (1989).

There is a considerable difference in the strength and thickness of fresh bird skins. The auk and crow families, for example, are very robust whilst others such as thrushes, doves and pigeons, are extremely delicate. This is important to consider when selecting skins for educational purposes, where handling may be an important element.

Cleaning and degreasing fresh skins

Most preparators do not clean skins routinely due to the danger of damage and/or colour change. It is good practice to clean when necessary either to reveal identifying characteristics or remove material which might cause long-term damage to the skin. The standard technique is to remove blood and dirt by swabbing the soiled area with water (see Primary specimen care, p. 1) or by soaking the entire skin in washing solution. This may be a dilute non-ionic detergent or, as suggested by Horie (1988), a more complex combination of chemicals and detergent. Whatever is used, it must be completely removed by rinsing with cold water.

It is important to remove as much fat as practicable from the inside of the skin. Its subsequent seepage and oxidation (fat burn) is one of the most common causes of bird skin deterioration. This is a particular problem with flat skins which easily lose feathers around the cut edges. File cards mounted on a wooden block or small wire brushes are efficient tools for this purpose. They may be used in conjunction with an absorbent powder such as borax, magnesium carbonate or cornmeal.

A variety of chemicals have been used to remove the remaining traces of fats and oils from the skin. These include dilute ammonia. chlorinated solvents, white spirit, petrol and industrial methylated spirit, although the last item has been specifically shown by Fry (1985) to affect the colour of some plumage (see also Colour changes in fur and feathers, p. 1). The ubiquitous 1,1,1-trichloroethane, favoured

because of its non-flammability, is no longer manufactured in most European countries because of health and safety considerations. Several substitutes have come on to the market, but are very costly and require further evaluation. Gerwin (1989) finds hexane to be an effective degreasing agent whilst Rogers and Daley (1988) suggests abandoning solvents altogether in favour of detergent solutions. They maintain that a small quantity of fat remaining in skin will cause little damage, and this should be traded off against the potential hazards of using solvents.

If washing and/or solvent cleaning has been used, the feathers can be brought back to their original condition (fluffing) by shaking in heavy magnesium carbonate or hardwood sawdust. The excess can be removed by means of a jet of compressed air directed from head to tail. This process must be carried out in a fume cupboard or dust-retaining box. Horie (1988) suggests that magnesium carbonate remaining in the feathers may have an advantageous buffering action by reacting with acids in the atmosphere to form moderately stable salts. Its disadvantage, according to Garrett (1989a), is that it may obscure details of feather follicles in flat skins. Hardwood sawdust is effective but also acidic, and may remain in the skin. For these reasons many preparators prefer not to use any powder cleaners, and simply dry the skin with a hair-dryer on a medium heat setting.

Traditional round skin preparation

In order to position the scapular feathers neatly, Chapin (1940) recommended stitching the feather tracks in a figure of eight pattern on the inside of the skin. An alternative method is to tie the humerus bones their normal width apart, at the elbow. This latter method has the added advantage of keeping the specimen together if it later disintegrates. The skin may be filled with a variety of materials such as cotton wool, chopped tow, polyester batting and wood fibre (wood wool). It may also be supported with an internal wooden stick or wire. In the case of small and medium sized birds, this internal support is allowed to project from the rear end of the skin, where it can be used for handling and as a support for the crossed legs. Labels are tied with thread either around the stick and legs or, in the case of wire supports, on a projecting wire loop.

Cato (1986) suggests using the preparation technique advocated by Harrison and Cowles (1970), which results in skins with flat backs. This will prevent them rolling about in collection drawers, thus causing feather damage. Flattened backs can also be achieved by gently compressing the skins whilst drying. Preparators use this technique routinely with larger skins to save on storage space. Conversely, some researchers insist on skins with rounded backs, as they more closely represent the living bird. Hangay and Dingley (1985) illustrate a wire-mesh drying rack specifically designed to achieve this end.

Before drying, the skin is wrapped with either a thin sheet of cotton wool or a length

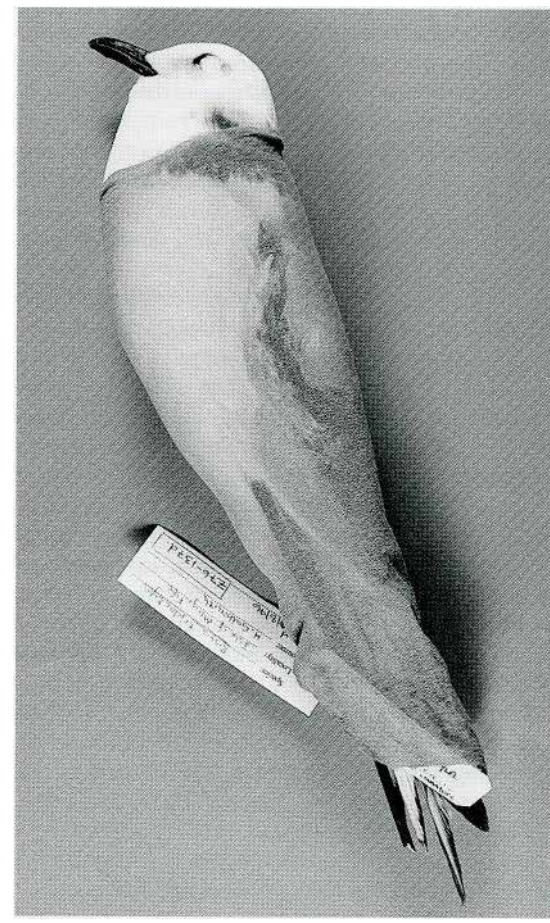


Figure 1.1 A nylon stocking can provide even compression of round bird skins whilst they are drying.

of paper pinned around the breast. Larger specimens can be inserted into cut lengths of nylon stocking which will evenly compress the skin (Fig. 1.1). Many European preparators completely wrap their skins in wet tissue paper. This results in a firm round skin but limits the opportunity to inspect the specimen regularly and correct distortion during drying. The bill may be held closed with a ball of softened wax or with pins. Van Tyne (1952) and Johnson *et al.* (1984) warn against sewing the bill closed with a thread through the nostrils, as this can lead to inaccurate bill depth measurements.

Variations in the basic round skin technique for dealing with larger birds such as herons are well illustrated in Anderson (1965), and are further discussed in Harrison and Cowles (1970).

Non-traditional skins/skeletons or combination specimens

Traditional round skins satisfied most research requirements for many years. In practice, most of the bird was discarded in favour of the well prepared skin. Barlow and Flood (1983) maintain that non-traditional skins were initially introduced to fulfil the needs of specific research projects. Norris (1961) is usually credited with starting this trend. His technique of gluing flat skins to card suited his particular requirements but, as a general method, it does limit access to some parts of the bird.

Increased communication between preparators, collections managers and researchers over the past ten years has fuelled a more general interest in preserving as much of the individual specimen as possible, but there is no consensus and techniques are still being debated.

Since museums have begun routinely to preserve both skin and skeleton (Spaw, 1989, calls these combination specimens) good illustrated accounts such as Rogers and Wood (1989) have become available. Apart from the flat skin with complete skeleton, the main variations are:

- Round skin with one wing removed and spread.
- Round skin with one boned wing and one boned leg, bill and skull remain with separate skeleton, one boned wing spread.

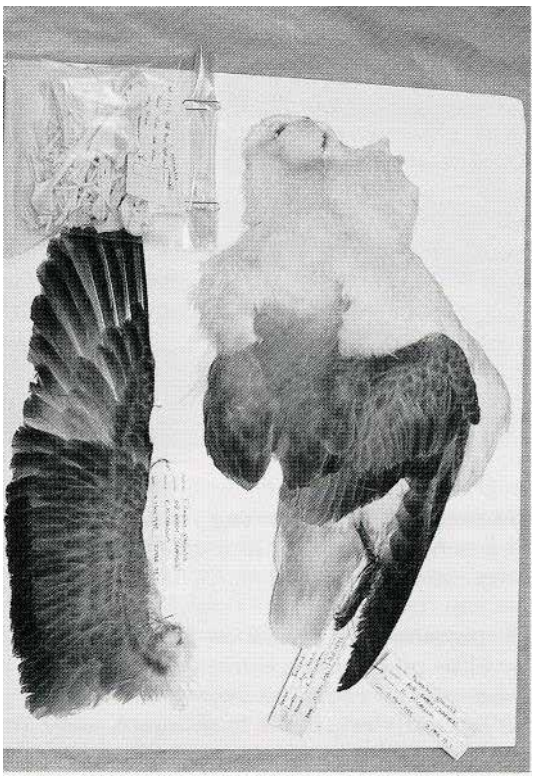


Figure 1.2 A non-traditional bird skin preparation which also preserves the whole skeleton.

- Flat skin with one boned wing and one boned leg, bill and leg remain with skeleton, one honed wing spread (Fig. 1.2).
- Complete skeleton with both boned wings spread.

The advantages of these variations is that they provide more information from a single specimen than traditional skins. They can be quicker and easier to prepare, and take up less storage space. Researchers will not, however, be able to check the standard measurements on a flat skin or those prepared without bills.

Preparation of mammal skins

Skinning

There are various ways to skin a mammal depending on the end use of the skin. Most are illustrated in standard texts such as Anderson (1965), Anon. (1968), Wagstaffe and Fidler (1968), Hangay and Dingley (1985) and Morris and Wroot (1987).

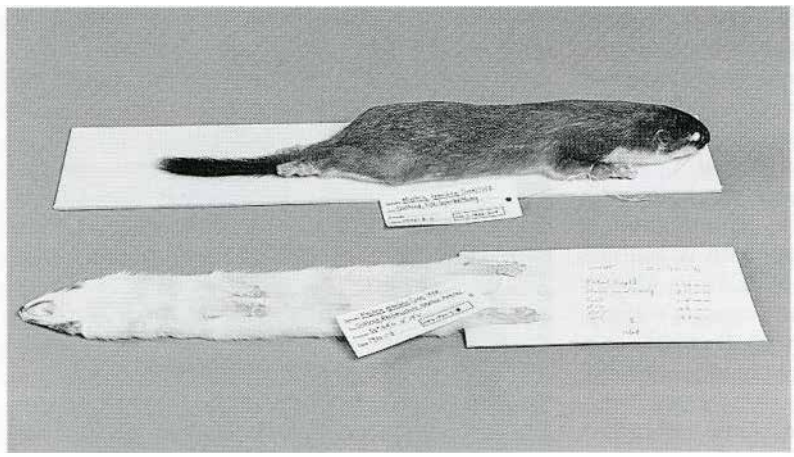


Figure 1.3 Round and flat (cased) mammal skins.

Cleaning and degreasing fresh skins
The same techniques are used as for birds (see Preparation of bird skins, p. 5).

Preparation of round or `study' skins Small to medium sized mammals may also be preserved as round or study skins (Fig. 1.3). An artificial body made from cotton wool, tow or synthetic fibre is inserted into the skin to provide both support and give it the approximate shape of the original body.

Supports inside the tail include wire (Nagorsen and Peterson, 1980, explain how to taper it), sharpened bamboo splints or stripped feather quills. A more flexible insert is thought to make the tail less susceptible to handling damage. Hangay and Dingley (1985) describe a method using silicone rubber sealant to fill the tail, but caution that the technique still requires evaluation.

Preparing symmetrical round skins requires skill and practice, but many researchers prefer working with them.

Preparation of flat or `card' skins
Small to medium sized mammals can be prepared as flat or card skins (Fig. 1.3) (also referred to as `cased skins' because of the skinning technique). The skin, either preserved or unpreserved, is stretched over a shaped insert made of card, plastic, wood or wire and then air-dried. If the card or plastic remains inside the skin, it may double as both support and label.

Two variations of this technique are also practised. Either both fore and both hind limb bones from elbow and knee remain in the skin and help support the fragile limbs, or the fore limb on one side and hind limb on the other are removed and preserved with the rest of the skeleton. The latter method has been adopted more recently, and reflects a general trend to make more of the skeleton available for study. In both methods the tail and legs are tied to the card with thread or fuse wire. Also favoured is the technique which involves the positioning of the limbs and ears of smaller mammals (i.e. fore feet glued in place, one palm up, the other palm clown and ears glued one forward, one backward) so that both surfaces of the ear can be examined.

The thin skins of rabbits and hares require special treatment. Dill (1950) and Thietje and Schrimpter (1967) use a solution of 50% industrial methylated spirit and 50% turpentine to preserve the skin. Anderson (1965) recommends using stout wires from fore to hind limb for support before inserting a thick card and wooden rod on which to tie the hind legs. A slightly modified version of this method, which involves removing a fore and hind limb for skeletal preparation, is used by Nagorsen and Peterson (1980).

Advantages of flat skins lie in their speed and ease of preparation, reduced storage requirements, secure labels, and the opportunity for retaining an almost complete skeleton. Disadvantages are possible hair loss through handling where the skin meets the card and, if the specimen is not properly prepared, loss of data through fat staining on the insert.

As a word of caution, some researchers have found it difficult to compare the pelage of flat skins with those prepared in the round.

Tanned mammal skins

In museum literature, tanning is often used as the generic term for the process of preserving mammal skins in a flexible condition. In the commercial tanning industry, the word tanning is reserved for the process which produces flexible, dehaired leather, and flexible skins with hair are said to be 'dressed'.

In the past, many skins were tanned with alum and saltpetre or salt and sulphuric acid. Although, as Reid (1985) explains, the detrimental effect of sulphuric acid on mammal skins was reported as early as 1907, it was still recommended in standard taxidermy texts until the late 1970s. The work of Hanacziwskyj *et al.* (1991) is the latest to confirm the potential long-term problem with this acid. In the last 20 years dressing agents such as Lutan F (aluminium chloride), together with organic acids, have become a safer and more durable alternative.

All dressing techniques are invasive and liable to increase the deterioration of the protein in the skin. Florian (1986) suggests that skin dressing may be unnecessary and has more to do with ease of handling large skins than with their longevity.

Handling and packing bird and mammal skins

Old collections in regular use should be tested for levels of harmful chemicals using spot tests such as those advocated by Found and Helwig (1995), and workers should be supplied with the relevant protective equipment. It is recommended that gloves are worn when handling any skin collection, both for the protection of the handler and the specimen. If the skin is stored in polythene tubing, it should only be removed for essential information which cannot be obtained otherwise. The long-term storage of skins in the same polythene tubing cannot be recognized as best practice. Polythene can degrade over time and, apart from splitting, the degradation products (which may appear as stickiness) can damage skins. It is recognized. however, that the protection afforded by this cheap and readily available material may be a trade-off against deleterious long-term effects.

Whenever possible, bird skins should be handled by their supporting sticks or wires and special care taken with vulnerable areas such as the head and neck of birds and the ears, tails and feet of mammal skins. Researchers should also be made aware of any particular problems with different collections. Some preparators, for example, sew the wings of round skins tightly to the body and the tails to the sticks.

Garrett (1989a) points out, from his observations, that flat bird skins stand up to handling at least as well as traditional round ones.

Loans of prepared bird and mammal skins from the reference collections are usually packed for transit in either stout cardboard or wooden boxes. All materials used in such packing should be of conservation quality. Previously used packing should be carefully examined for signs of infestation, dirt or weaknesses before re-use. It is recommended that small to medium sized skins should be wrapped in tissue paper and packed in layers of cotton wool or polyester batting. Some institutions also insert skins into cardboard tubes as an extra precaution. Hinshaw (1989) gives a detailed account of packing traditional bird skins and skeletons. Her methods can be adapted for most types of mammal skins.

Osteological preparation techniques

Museum osteological collections have suffered in the past because of tradition and practicability. The traditional round skin was the favoured component of most bird research collections. The skeletal elements retained were beyond easy reach, inside the skin. In the case of mammal collections, it was usually only the skull which was retained with the skin. To some extent, this reflected the resource problems associated with preparing and transporting dry vertebrate material. If you have post-cranial material more than ten years old it is the exception, and should be valued as such.

The lack of fish and reptile osteological material in museum collections is to some extent due to the difficulty of preparing these groups.

Vertebrate skeletons are composed of bone and cartilage. Immature skeletons and those of sharks, rays and some amphibians are mostly or wholly composed of cartilage. As cartilage is prone to shrink and distort on drying. these groups are usually preserved as wet preparations. Wagstaffe and Fidler (1968) recommend formalin for their storage, whilst Mahoney (1973) suggests that they may be hardened in formo alcohol and stored in industrial methylated spirit. Knudsen (1966) describes in detail the preparation of cartilaginous fish skeletons (see also Chapter 5 on Fluid preservation).

There are only a limited number of methods for preparing vertebrate skeletons, but individual variations are considerable. Historically, the search for the ideal technique was based on speed and good results. Today this list would be headed with the technique which would result in the least long-term damage.

It is essential for any preparator to become familiar with the vertebrate skeleton through anatomical texts such as Sissons and Grossman (1975), Ellenberger *et al.* (1956) and Saunders and Manton (1967), and from previously prepared material. Unfamiliarity will lead to loss or damage of bones (Fig. 1.4). Behrmann (1979), when dissecting three whales, reported the anatomy to be revealed in the first. considered in the second and explored in the third — a methodology to be recommended for all unfamiliar groups. It is also essential to identify material correctly before it is skeletonized, and to document reasons for the identification.

The end use should be determined before a skeleton is prepared. Research collections demand either complete disarticulation (Matthiesen, 1989, considers this to be essential for bird collections) or partial disarticulation (ligamentary skeletons). Display exhibits are usually ligamentary or fully articulated. Thompsett (1958), Adams (1980) and Hangay and Dingley (1985) describe techniques for articulating skeletons, and Behrmann (1979) illustrates the ten most commonly observed errors in articulated cetacean skeletons.

The bones and cartilage of embryos, small fish, amphibians, reptiles and small mammals may also be prepared as stained preparations and later embedded in clear plastic for display and handling collections. These techniques are fully described in Hangay and Dingley (1985).

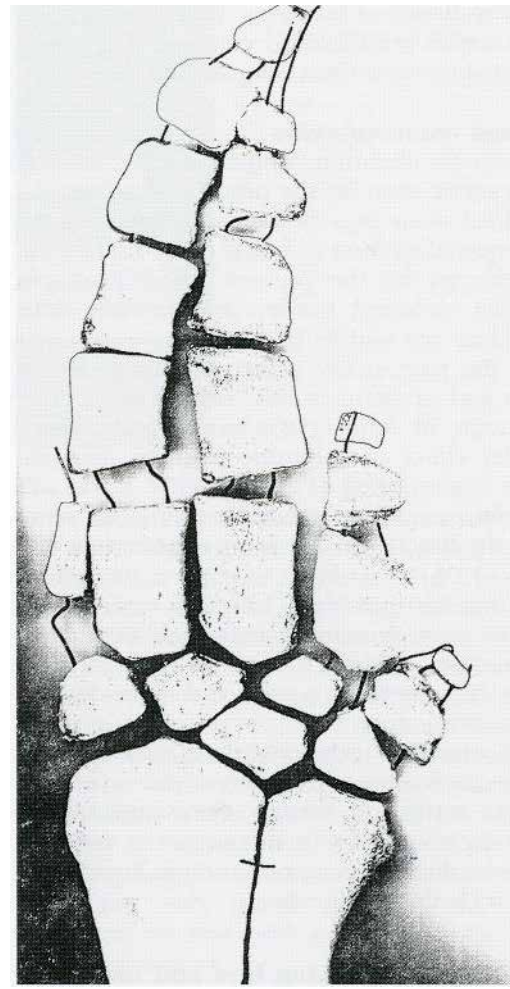


Figure 1.4 A photocopy of a dolphin 1 rib used as a reference for rearticulation purposes.

Labels for osteological work must be made of a material that will survive the preparation processes. Matthiesen (1989) prefers embossing draft Mylar film, whilst Davis and Payne (1992) use the same technique with aluminium foil (Fig. 1.5). Tyvek may also be permanently embossed with a hard instrument. A reusable alternative reported by Trodd (1993) is to use numbered lead tags.

Dissection

Ligamentary articulated skeletons of fish may be prepared by simple dissection. Konnerth (1965) describes the technique and maintains that it works best with pre-frozen material.

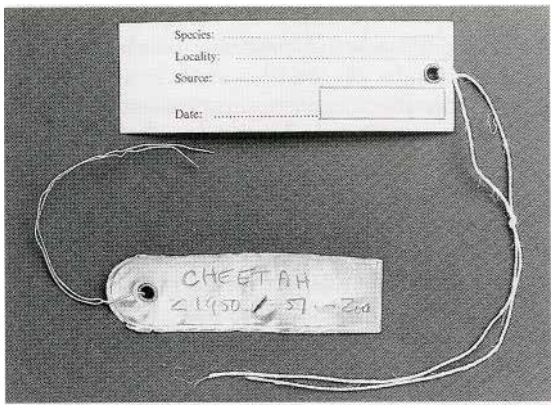


Figure 1.5 Tyvek and aluminium labels.

Burial

This technique is probably the oldest method known and, with very large specimens, perhaps the only practicable solution, but it is slow. Adams (1980) found that an elephant skull took up to two years. Moreover, it is difficult to monitor and may result in significant discolouration of the bones.

A variety of burying media have been suggested. Scharff (1911) worked with a large pit of beach sand, whereas Adams (1980) prefers finer silver sand. Hounsome (1988) suggests that mixing soil with sand will speed up the process and Hendry (1993) discovered a museum in North America using cow manure! Davis and Payne (1992) have achieved good results with well rotted leaf manure, burying roughly fleshed carcasses in bags made from parachute silk.

Cold water maceration

This is commonly used to produce ligamentary skeletons for display and teaching purposes from roughly fleshed material. Thompsett (1958) warns that if preliminary fleshing is not thorough, adipocere (an insoluble by-product of the decomposition of fat) may be formed during subsequent cleaning and this is very difficult to remove.

Although slow and very smelly, cold water maceration is easy to monitor and very suitable for immature and cartilaginous skeletons. The smell can be reduced by using running water, frequently changing the water or by using agar, as recommended by Hurlin (1918). Wiles (1932) found that agar could also be used for gently disarticulating mammal skull bones.

Before commencing any macerating procedure the long bones of mammals may be drilled to facilitate the removal of fat. Care must be taken, however, to avoid damage to diagnostic features. After maceration, fat removal can be assisted with jets of water or compressed air.

Matthiesen (1989) uses cold water maceration to produce completely disarticulated postcranial bird skeletons after preliminary treatment in a dermestarium (described below). Final cleaning takes place in a heated ultrasonic bath containing water and detergent, a technique also recommended by Spence and Tonkinson (1969).

Glass jars and fish tanks are ideal for smaller specimens. For medium to large material Adams (1980) used a plastic water butt fitted with a filter trap and hose pipes to service the process hygienically.

Warm water maceration/simmering

This is a popular technique because of its speed, but it is still a smelly operation. Before proceeding, the brain should be broken up and removed either by suction or syringing with a stream of cold water. This will prevent it swelling during maceration and damaging the elements of the skull. Coy (1980) also slits the gums of mammals to prevent teeth being pulled out as the gum shrinks.

Adams (1980) uses macerating temperatures ranging from 37 to 80°C depending on the material being processed. Thompsett (1958) insists that proper cleaning can only be achieved by simmering between 60 and 65°C in order to destroy the periosteum. Most preparators caution that the use of prolonged boiling will damage osteological material.

Stainless steel or aluminium containers are commonly used with this method. They may be fitted with a grill to lift bones out of the sediment and to prevent damage if the container is left to evaporate completely.

Chemical maceration

Mahoney (1973) and Hangay and Dingley (1985) describe the traditional technique of adding a small quantity of washing soda to bones simmering in water. This may also help remove some of the fats.

Sodium perborate was originally used by Roche (1954) at the Natural History Museum in Paris, and introduced to the UK in separate papers by Chapman and Chapman (1969) and Coy (1975). Coy's description purports to follow closely the original method where boiling water is poured on to the bones and dry perborate before the container is sealed. A popular variation is simply to add a little sodium perborate to material simmering in water. Flesh remaining on the bones turns to a jelly-like consistency, and is quite easily removed. This technique considerably reduces the smell associated with osteological work. Davis and Payne (1992) insist that the use of sodium perborate must be carefully monitored as it can result in bones becoming soft and chalky.

Hangay and Dingley (1985) describe a technique for preparing adult skulls using sodium bicarbonate, ammonia and sodium hypochlorite. Although quick, the skulls are subjected to prolonged boiling, a treatment which Williams and Smith (1995) regard as the most destructive of osteological techniques.

Green (1934), Harris (1959), Mahoney (1973) and Hangay and Dingley (1985) describe techniques using antiformin, a solution of sodium carbonate and calcium hypochlorite, for preparing material previously preserved in alcohol or formalin (see also Chapter 5 on Fluid preservation).

Enzyme maceration

A variety of enzymes are used for skeletal preparation. Shelton and Buckley (1990), however, have pointed out that there has been little research on their effects on skeletal material and, although they are included here to demonstrate the range of techniques commonly used, enzyme maceration may limit a collection's future research potential.

Moyer (1979) and Fisher and McInnes (1981) have both used pancreatin (Rowley's fluid), and used it for both skulls and post- cranial elements.

Harris (1959) recommended normal saline for completely disarticulated skeletons, and Mahoney (1973) notes that this is also useful for preparing bones from owl pellets. Mahoney (1973) also suggests trypsin in sodium carbonate for general osteological work, whilst Piechocki (1986) prefers pepsin on its own. A relatively new enzyme is neutrase, a bacterial protease produced by a selected strain of *Bacillus subtilis* and widely used in the food and brewing industries. Davis and Payne (1992) are its main advocates and describe techniques for its use.

A range of biological detergents has also been used and Howard (pers. comm., 1996) finds that Persil is the most effective. The inclusion of sodium perborate in this product may contribute to this conclusion.

All enzyme methods require the bones to he incubated at temperatures ranging from 37 to 50°C. They are quick but invasive and very smelly. Shelton and Buckley (1990) caution that denaturing the enzyme at the end of the process is problematic and may itself lead to hone damage.

Invertebrates

Both adults and larvae of invertebrates have been used to prepare vertebrate skeletons. Bolin (1935) describes a technique for preparing fish skeletons using marine isopod species, and Hounsome (1988) has carried out osteological experiments with freshwater ostracods. Banta (1961) lists variety of arthropods used for vertebrate skeletal preparation, and describes his own work with clothes moth larvae (Tineidae species; see Chapter 8 on Pest management, prevention and control). Both Allen and Neill (1950) and Ilangay and Dingley (1985) have experimented with mealworms *(Tenebrio ntolitor* or *T. obscuruis).*

Dermestid beetles are the most widely used, and probably the most convenient for museum use. Laurie and Hill (1951) report that the smallest immature bat skull can be cleaned without sutures opening or teeth falling out. Williams (1992) recommends dermestaria together with vacuum cleaning as the least invasive technique to prepare osteological material for research collections. This avoids the need to soak such material in water which Williams (1992) has found to damage mammal teeth. Williams and Smith (1995) also suggest combinations of these three techniques may have an effect on the dimensions of mammal skulls. Using invertebrates such as dermestid beetles may be the only technique which can he recommended from a long-term conservation standpoint. However, the location of the

laboratory, frequency of preparation and allergenic reactions to dermestids also have to be considered.

There is extensive literature on the construction and management of dermestaria, and it stresses the importance of the correct environmental conditions for the survival of the colonies. The best recent accounts are those by Storer (1988) and Matthiesen (1989) which also detail instructions for disarticulating and fleshing birds for such colonies. Anderson (1965) and Hangay and Dingley (1985) provide similar instructions for disarticulating mammals.

Hendry (1993) reports that museums may use up to five species of dermestid beetles in mixed colonies, a practice which Hounsome (1988) recommends as it takes advantage of the different size, food preferences and optimal living conditions of the insects. If there is concern over the location of the colony within a museum, Marcon (1992) gives specifications for a transportable dermestid colony building with environmental controls. Willard (1989) suggests that pheromone traps could be employed in the rooms adjacent to dermestaria for trapping escapee beetles (see also Chapter 8 on Pest management, prevention and control).

Previously preserved skulls are not always palatable to beetles. De la Torre (1951) suggests coating them with bacon fat, whilst Hooper (1956) prefers to apply cod liver oil. Storer (1988) simply soaks skulls in ammonia overnight before treatment. Very dessicated skulls were successfully cleaned by Laurie and Hill (1951) after soaking in Marmite (a vegetable extract). Sommer and Anderson (1974) maintain that the unpalatability of formalin-treated specimens can be used to advantage when preparing ligamentary skeletons. Brushing formalin on to joints ensures that they are not eaten and so do not disarticulate.

After cleaning, a dilute solution of ammonia in water (nine parts to one) can be used to remove remaining grease and, or dermestids. Freezing is also commonly used for pest control, but Williams (1992) suggests that a period of quarantine and observation is the least invasive technique.

Degreasing

It is necessary to remove most of the fats and oils from osteological material. The oxidation of large quantities of leaked fat and the subsequent accumulation of dirt can destroy diagnostic features, obscure labelling and make handling unpleasant. This process must be carefully monitored, as removing all the fat can lead to splitting of bone.

It is cetacean material and large mammal bones that are the most difficult to treat. Such specimens may require degreasing in a commercial vapour degreaser with an organic solvent (see anon., 1989). These are expensive to purchase and operate safely. However, most veterinary teaching institutions and some larger museums operate them, and may process material for the cost of the solvent.

Acetone, trichloroethane, white spirit and petrol have been used for degreasing bones. All have their own problems with respect to flammability, toxicity and availability, and should only be used strictly with reference to their data sheets and health and safety regulations. Matthiesen (1989) found Stoddard's solvent to be preferable to both carbon tetrachloride and acetone for bird skeletons.

Other degreasing agents include sodium hydroxide, used by Knudsen (1966) as a 2% solution and by Entwistle (1992), slightly stronger, at 4.5%. Hendry (1993) reported 10% ammonia being used in some North American museums, and concentrations of this chemical up to 20% have been adopted by Jannett and Davies (1989) for use with their skull degreasing apparatus. Although Anderson (1965) suggested degreasing bones in 5—10% aqueous sodium bicarbonate, he warned that this technique required careful monitoring to avoid damage. Matthiesen (1989) and Coy (1980), both advocates of the sodium perborate technique, maintain that with this method further degreasing is usually unnecessary.

Martin (1964) suggests covering bones with a cloth during degreasing. The cloth prevents fat coming into contact with the hones during removal of the material from the container.

Bleaching

This is not usually considered necessary for osteological research collections. Mitchell and Wynne Jones (1956) maintain that aqueous solutions of hydrogen peroxide, the most commonly used bleach, are acidic and that this may damage bone. Coy (1980) and Storer (1988) also caution against the use of peroxides.

If a bleach must be used, it should be reserved for exhibition specimens. Chloramine T has been suggested as a substitute for peroxides but it is known to be difficult to remove when used in paper conservation, where a very dilute solution of calcium hypochlorite is the recommended, less damaging, substitute.

Conversely, Davis and Payne (1992) stain some of their bone material with strong tea in order to accentuate detail under the microscope.

Specimen documentation and labelling

General procedures for documenting and cataloguing natural history collections are widely discussed in Davis (1994). Most institutions base their hard copy records on computer-friendly recording cards such as the generic MDA natural history card or more specialized in-house ones.

For information relating to specific dry vertebrate groups see Williams *et al.* (1977), Waddington and Rudkin (1986), Cato (1986), Genoways *et at* (1987) and Simmons (1987), and for documentation guidelines see Garrett (1989b).

Conservation records

The importance of recording conservation information and procedures has been stressed by most writers in this field and is detailed in Chapter 9 on Policies and procedures.

There is no universally set procedure but most workers agree that records begin with the death of the specimen. The cause of death, if known, should be recorded, together with any action or treatment which might affect the future research potential of the specimen.

A clear and simple form has the best chance of being completed. Its most important feature is the specimen reference number which links the specimen to the record card. This is especially important when parts of a specimen are divided into different storage areas in the collections.

Condition reports are mainly used for vertebrates on loan or for assessment surveys. Generic bird and mammal diagrams can speed up the process considerably (see Chapter 9 on Policies and procedures).

Labelling (see also Appendix II on Papers, inks and label conservation)

Specimen labels should never be removed unless their acidity is causing significant damage to the specimen or they are themselves disintegrating.

Tie-on labels are usually attached to the legs of both reference and mounted specimens or to the antlers and horns of game heads. Good quality linen and cotton thread are recommended. The latter is reputed to he less liable to rot in adverse conditions. It has become standard practice to loop the thread through the eyelet and knot it a short distance from the specimen. This allows comfortable access to the information without undue stress on the specimen. To avoid possible damage to the label, eyelets should be made of a non-corrosive material.

There is general agreement, supported by Hawks and Williams (1986a), that a good paper for dry collections labels is 100% cotton stock with a pH between 6.5 and 7.0.

Hendry (1993) reported that Tyvek (spun-bonded polyethylene), Byron Weston's Resistall 28# and 36# and Goatskin Parchment, used mainly for wet collections, also have their dry collections advocates, and can be useful for field-work and freezer labels. If Tyvek is used for tie-on labels it is advisable to protect the holes with non-corrosive eyelets. Sometimes this generally resilient material can be torn surprisingly easily with the tying thread.

Inks and pens *(see also Appendix II on Papers, inks and label conservation)*

There have been a series of research projects on the most suitable inks and pens for labels, store-boxes and polythene bags. Although inks are to be recommended for legibility, embossing labels may be a surer method of preserving documentation during some preparation techniques.

Osteological collections

Disarticulated osteological collections are usually numbered on the individual bones where practicable. The inks recommended are the same as for tie-on labels. Occasionally bones are resistant to ink because of the preparation technique, or it may bleed into porous areas. The MDA (1995) maintain that

brushing on a layer of PVA or Paraloid B72 and letting it dry will provide a surface suitable for labelling. The ink may be coated with a further layer of these materials to protect the documentation.

Organization and storage of collections

The best source for information on creating, managing and monitoring stores and selecting storage equipment is Moore and Williams (1995). Mathias (1994) and anon. (1992a) suggest useful guidelines for the organization of storage areas (see also Chapter 7 on the Collection environment).

Most dry vertebrate reference collections of skins, skeletons, nests and eggs are stored in taxonomic order and catalogued according to a widely recognized phylogenetic system. See Cato (1986) for bird systems and Williams *et al.* (1977) for mammals.

Mounted specimens and other dry vertebrate materials that have been collected or prepared especially for educational handling projects are usually stored separately from the reference collections. It can be difficult to store these collections in any way other than a notional phylogenetic system because of the differences in size and shape.

Scarce resources have focused debate on the general use of more expensive, conservation grade materials for the preparation and storage of all types of natural science material. Research is not yet available to settle these arguments and a pragmatic approach is usually taken by collections managers. Adopting higher standards of care for dry vertebrates, however, can only improve the general regard for these collections. Their low status has often been the root cause of neglect.

Traditional study skins

In collections with good standards of preventive conservation, traditionally prepared study skins may be stored in cabinets containing drawers lined with polyethylene foam or 100% virgin polyester felt. As a second level of protection or for cabinets which are not air-tight, they may be stored in resealable polythene tubing (Fig. 1.6). Mathias (1994) illustrates a variation of this method which ensures safe and easy access to the specimen. Small skins may be stored in zip-top polythene bags. Preparators must ensure that skins are as dry as possible before sealing in polythene. Air-drying is the safest method. Regularly monitoring such collections is vital and, as all skins retain some moisture from the atmosphere, there is the danger of a hostile micro-climate developing inside the sealed polythene. If this becomes a problem, one end of the tubing should be left unsealed.

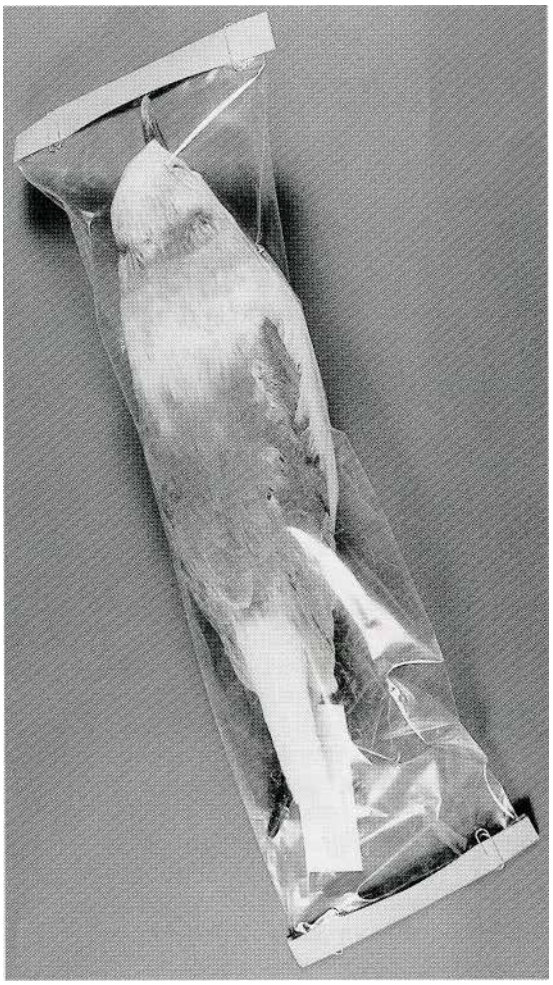


Figure 1.6 A round bird skin stored in polythene tubing sealed at each end. The risk of damage is limited as the skin may exit and enter the tubing with the lie of the feathers.

Flat skins

Flat bird skins

Large, flat bird skins may be folded and stored in polythene bags. Small and medium sized ones may require additional support. Garrett (1989a) uses sheets of heavyweight blotting paper, an easily replaced material which may absorb any excess fat from the skin. The skins are stored in unsealed polythene bags. Foam board is a useful alternative to blotting paper for heavier skins.

Tanned mammal skins

Hawks *et al.* (1984) is the standard text on tanned mammal skins. Traditionally, these skins have been hung from hooks in their eye openings. This can put considerable strain on the skin and it is recommended that all tanned skins, where practicable, are best stored on flat shelves. This does present difficulties for museums with large mammal skin collections, and Hawks *et al.* (1984) describe an alternative hanging system using polyethylene-foam covered supporting tubes and recommend an acceptable environment for cold room storage of between 20 and 22°C with a relative humidity (RH) of 50-60%, in conjunction with a fumigation programme.

Pool (1997) suggests raising the temperature of cold rooms to within 10°C below the surrounding room temperature, to help reduce condensation problems during specimen transfers but stresses the importance of also implementing an integrated pest management strategy, monitoring RH and fitting alarms.

Eggs and nests

Tennant and Baird (1984) have reported that birds eggs can suffer damage from the gases released by some wooden storage cabinets. The ideal solution is to store them in conservation quality card trays, card boxes with a Mylar window or polystyrene boxes, nestling them in 100% cotton. The use of metal cabinets is becoming universally recommended as best conservation practice. Kishinami (1992) illustrates a cotton poncho technique which eliminates movement of the eggs within the drawers during use.

If loose, nests may be bound with cotton twill tape and stored on trays inside cabinets.

Further protection, such as that suggested by Fuller *et al.* (1992), may be necessary if the collection is regularly used. It is useful to indicate on the cupboards the fragility of such material.

Storage of osteological collections

Where practicable all skeletal material should be housed in containers large enough to allow easy access without damage. Williams *et al.* (1977) recommended environmental conditions of 21°C and RH of 55% for this type of material.

Most small skeletons may be stored in glass vials, clear polystyrene boxes, conservation quality cardboard boxes or trays and translucent polyethylene/polypropylene boxes inside cabinets. Morgan (1991) points out that, whilst polystyrene is not prone to deterioration through oxidation, it may yellow and craze if

Figure 1.7 Separate parts of the same skeleton can be stored in polythene bags fastened together with plastic garment tags.

Figure 1.8 Tyvek-covered giraffes in the basement of The Natural History Museum, London.

exposed to sunlight. For further information on the choice of plastics for use in conservation see Baker (1995).

Matthiesen (1989) suggests zip-top plastic bags in clear plastic boxes as an economical alternative for avian skeletal material, and promotes storage on open metal shelving as a sensible way of servicing such collections. Separate parts of the same skeleton may be stored together in polythene bags held together with a nylon tag (Fig. 1.7). This system is useful for a variety of purposes, including physically separating specimen and documentation in cases where the acidity of the label could cause damage.

Medium to large skeletons may be stored in conservation quality cardboard boxes or hand-made polypropylene fluted-sheet boxes. Tetreault and Williams (1992) and Schlichting (1994) describe methods for preparing the plastic variety. Techniques for housing, cushioning and supporting fragile material can be found in Rose and de Torres (1992). Large skulls with horns or antlers attached to shields may be stored in the same way as game heads or separately on shelves. Of all the materials used to protect dried vertebrate specimens from dust, Tyvek covers, being simple to make and easy to clean, are most to be recommended (Fig. 1.8).

Figure 1.9 Purpose-built mobile cradles made of angle iron allow large cetacean skeletons to be moved by one person.

Large bones can be very susceptible to attack by insects, as they usually contain some residual fats and oils. Hendry (1993) mentions a museum with this particular problem having to seal specimens into wooden crates using a glue gun on the lids. If you have to use glue guns it has been recommended (anon., 1992b) that the safest glues from a conservation standpoint are 3M 3764, Bostik 6363 and Evostik 7702.

Even the largest cetacean skeletons can be made manageable on custom-built cradles of angle iron (Fig. 1.9) and polyethylene foam or of metal and nylon webbing, as illustrated in Potter and Heyning (1992).

Public display and teaching collections

Taxidermy

When commissioning taxidermy work for exhibition or teaching, it is important to obtain good quality specimens mounted by profes-

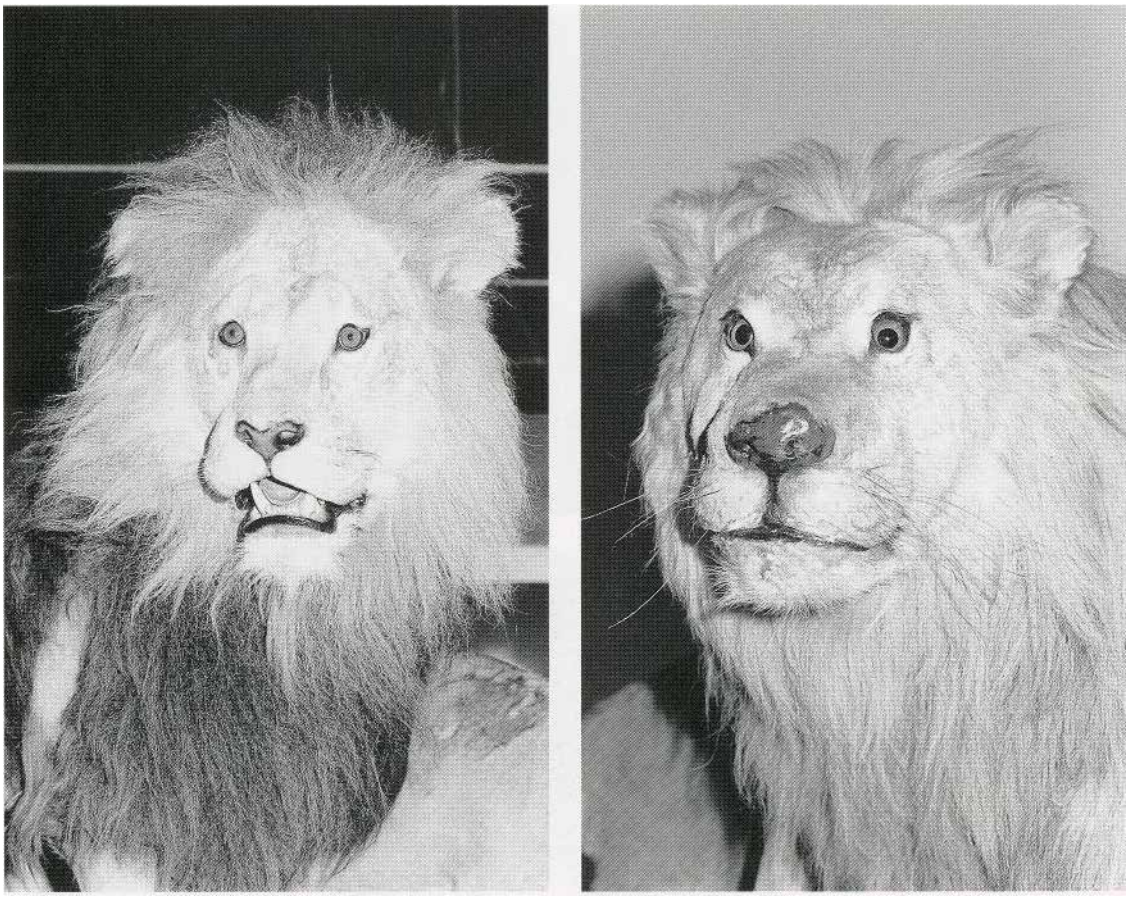


Figure 1.10 Good (a) id had (b) taxidermy is not always as obvious as with the two mounted lions. eking professional advice is good practice.

sional taxidermists. Judging the quality of a specimen requires experience and, if in doubt, seek professional advice (Figs 1.10a and b). The best taxidermy will never be cheap, but it will attract an audience beyond the purely educational.

Birds

The earliest surviving mounted birds contained much of the skeleton supported by an internal wire framework. Illustrated accounts of early work can be found in Bullock (1817), Brown (1885) and Hornaday (1921). Specimens were filled with a variety of soft fillings such as sawdust, straw, cotton and tow. Gardner (1880) points out that variations in this basic technique were often named after their proponents. Morris (1983, 1986) has found this to be useful when dating such specimens by X-ray analysis. Additional dating evidence may come from investigating the glass eyes (Fig. 1.11), which Gutebier (1987) and Mildner (1988) show to have undergone evolution in terms of their manufacture. Eventually, the method of skinning a bird leaving the skull, leg and wing bones attached to the skin and wiring them into an artificial body became standard practice. This is basically the technique favoured by taxidermists today. The most popular material for bird bodies has long been wood wool (chopped wood fibre) although carved balsawood and polyurethane foam are often used today.

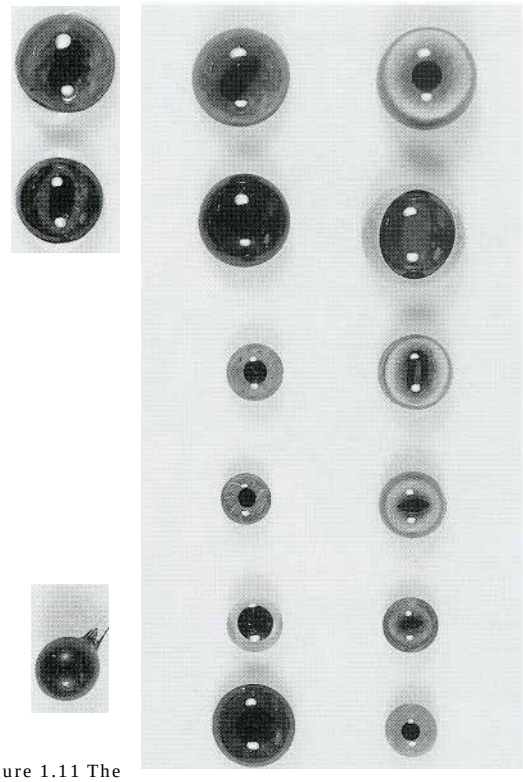


Figure 1.11 The type of glass eye used may provide useful information when dating specimens.

Mammals

The evolution of mammal taxidermy methods is not so simple. Early attempts mirrored those used for birds; Akeley (1923) recounts that the preserved skin was wired and literally stuffed with soft filling. Reports on the 1851 Crystal Palace Exhibition (anon., 1982) show that a variety of methods were discovered, lost and rediscovered by the subsequent generation. At the end of the nineteenth century, standard techniques had been developed that are still used by taxidermists today, albeit with new materials.

Small and medium sized mammals were usually prepared by binding their wired limb bones, skull and tail with wood wool to replace the muscles. Chopped wood wool was used to fill the skin and, although the technique sounds primitive, in the hands of a skilled and knowledgeable taxidermist, it produced very credible results and is still practised today.

Most large mammals have been mounted by variations of two basic techniques: direct modelling and the dermoplastic or Akeley method.

1. Direct modelling

This is the older method, and involved using the cleaned limb bones, skull and pelvis. These were attached to a wooden backboard with bent metal rods which supported the whole armature. Wood wool, bound to this framework, produced the body and muscle shape (Plate 3), and a layer of papier middle was applied to smooth contours and add detail. Several layers of shellac rendered the papier mache waterproof. The preserved skin was either pickled in a chemical bath or commercially dressed, before being glued and sewn around this mannikin.

Mammals such as elephants and giraffes mounted by this technique can present real problems with handling and floor loading as well as skin splitting. Hendry (1989) reported that experiments had been carried out to remove the entire framework of these large animals through an opening in their abdomen, replacing the support with fibreglass and polyester resin (Fig. 1.12). Although successful, it will take time to monitor the long-term effects of the fibreglass laminate on the skin.

2. Dermoplastic or Akeley method

This technique is justly named after its foremost American proponent, the taxidermist Carl Akeley – although, for the record, similar methods were used much earlier and with considerable success by taxidermists in Europe such as Karl Kusthadt and Leopold Martin (illustrated by Jahn, 1995).

The armature was prepared in a similar way to the direct modelling method, although the whole of the skeleton was sometimes used to achieve greater accuracy. Modelling clay created every detail of the body form (Fig. 1.13). The clay model was moulded in plaster of Paris and cast in plaster, reinforced with hessian. There were many variations of this technique, especially the use of glue and paper to prepare the cast. The result was a strong, lightweight, hollow mannikin on which the skin was mounted (Fig. 1.14).

Today both techniques are still in use but the Akeley method is the one favoured by the

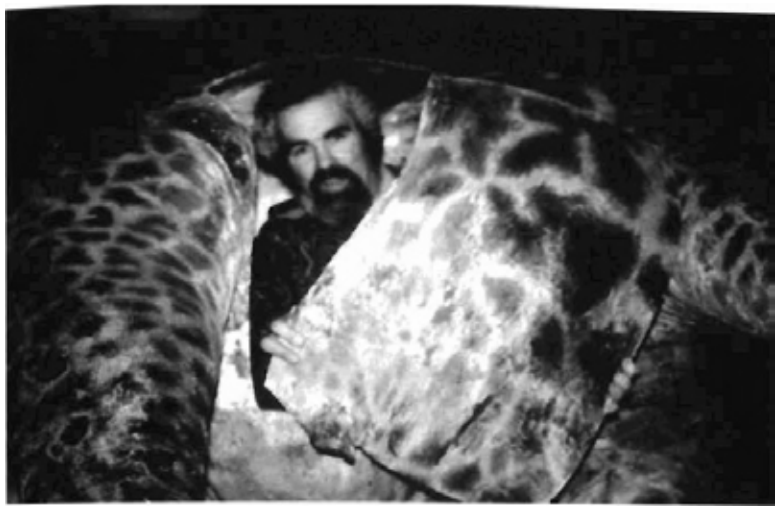


Figure 1.12 The most radical technique to ease handling of large mammals is to replace the heavy internal armature with fibreglass.

Figure 1.13 The clay maquette technique is used to assess the stance and muscle structure before embarking on the life-size mannikin.

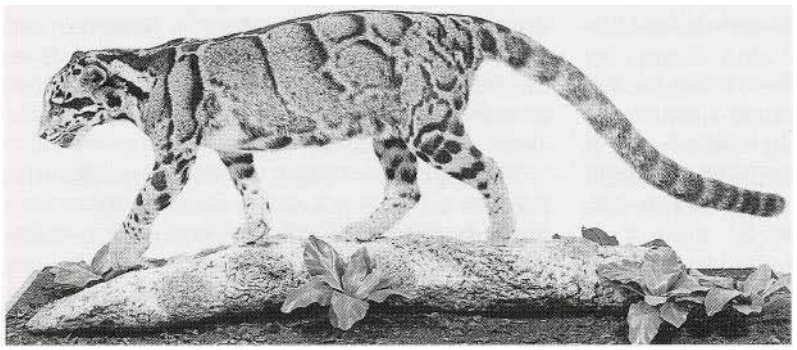


Figure 1.14 A clouded leopard mounted by the Akeley method.

leading proponents and can result in exceptional examples of the taxidermist's art. Jonas (1930) describes a more sophisticated variation of this technique used to mount elephants.

Commercially produced mannikins made from paper or polyurethane foam are now available for almost every common mammal from weasel to polar bear. Both polyester and epoxy resins have also been used in large mammal taxidermy by museum and commercial taxidermists for over twenty years. We should be concerned that materials such as polyurethane foam and modern resins, which form the internal support for so many museum specimens today, have been used without the benefit of any published research on their long-term suitability for this purpose.

The sparsely haired, thin skin on the hands, feet and faces of mammals such as the large apes has always been a problem for taxidermists. Attempts to achieve the correct translucency together with minimal shrinkage led to the development of a technique, reported by Kaestner (1959), for replacing the skin with an artificial material.

The raw skin of a specimen was fitted over a clay-covered armature and modelled under the skin to a finished state. Wax was sprayed over the fur followed by a plaster jacket in sections. The skin was allowed to decay in a warm water bath, leaving the hair embedded in the wax. The skin was replaced with coloured wax, and the mould eventually removed (Plate 4). The resulting mounted specimen consisted of wax and hair only. In recent years taxidermists such as Mayer (1987) and Epping and Epping (1981) have introduced modern rubbers and resins as a more permanent substitute for wax.

Specimens produced by this method, as well as being extremely lifelike, are very resilient, as they do not have the problems of skin shrinkage or infestation usually associated with a traditional skin mount. As a method, however, it requires considerable skill, practice, ingenuity and time, and is only one step away from producing lifelike models in place of real specimens. Indeed, the work of Kung (1982) has shown that it is possible to produce convincing artificial models of rare species for museum exhibition. However, they lack the charisma of real specimens.

Fish, amphibia and reptiles

All members of these groups can be mounted by traditional taxidermy techniques using the original skin. In many cases the resulting specimens are difficult to handle because of their fragility. Also, because of the technical difficulties of capturing the translucency and depth of colour of the skin, they may be of only moderate quality. For display or teaching purposes, it is usually preferable to make whole or partial reproductions (Plate 5). Migdalski (1981) illustrates and describes several techniques for reproducing fish.

There is a considerable history attached to moulding and casting techniques with this group. Reproductions in museum collections such as those by Walters (1925) have a value in themselves as part of this history.

Freeze-drying

Freeze-drying is a method of preserving natural history material by slowly extracting the water from frozen specimens. Whole specimens may be freeze-dried (Fig. 1.15) and, unlike traditional taxidermy, no skinning may be necessary. There are, however, many variations of freeze-drying methods, some including more traditional taxidermy techniques.

Freeze-drying is usually carried out in commercially available freeze-dryers, although home-made systems have been described by Kelly (1980). The basic unit consists of a deep-freeze chamber in which a vacuum can be maintained attached to a separate condenser. Under such conditions sublimation of the ice within the specimen to vapour takes place. This vapour is removed and refrozen, some distance from the specimen, on the condenser. As the cellular structure at any point in time is either frozen or dry, shrinkage is considerably reduced when compared to normal drying. Eventually, all the ice is removed and although the specimen, once removed from the freeze-dryer, will absorb a certain amount of water from the atmosphere, this is insufficient to cause decay.

Although, as Sage (1984) reports, the process has been known since 1890, it was not until the 1960s that equipment became available for use with whole specimens. Museums became familiar with the technique through the work of Meryman (1960, 1961),

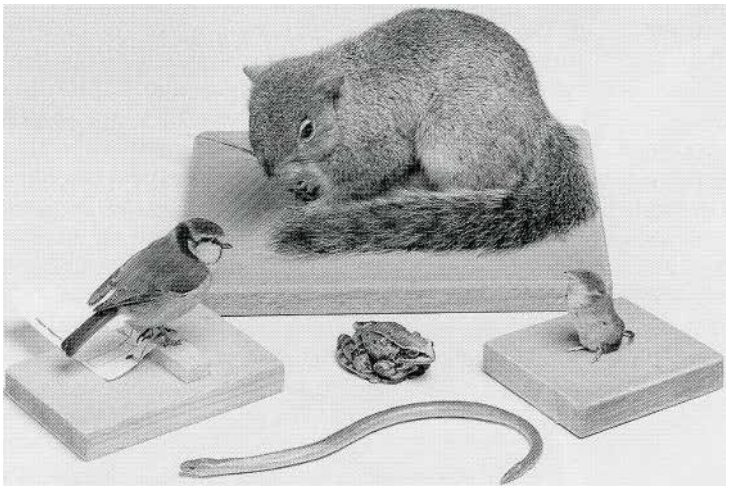


Figure 1.15 Some examples of freeze-dried specimens.

Harris (1964) and Hower (1979). Although freeze-dryers are still expensive to buy and maintain, their reliability has improved considerably relative to the earliest models.

Freeze-drying can be used to preserve all vertebrates but in practice its use is dictated by the size of specimens and their suitability for the process. Time is also an important element as even medium size entire specimens can take several weeks to dry. Reptiles, and to a lesser extent amphibia and fish, can be successfully freeze-dried, but the preparation is far more elaborate than for birds and mammals. The colour of birds' legs and bills fades with normal drying but with freeze-drying it can disappear completely, and they take on a bleached appearance. Hangay and Dingley (1985) give the best practical account of freeze-drying procedures and detail methods for all types of natural history material.

Freeze-drying is used mainly for display and teaching. The freeze-dried body provides its own internal support when dry, making it more robust than traditionally prepared specimens. It is also widely used for the dry preservation of decaying specimens, which would otherwise have to be preserved in fluid. Moreover, the skills required for preparing freeze-dried study skins are easily acquired, unlike the depth of knowledge and skills required of a trained taxidermist.

Smaller bird and mammal mounts can be prepared by setting up the entire specimen. However, evisceration and packing the body cavity with soft filling will reduce the drying time. The technique for larger mammals is to freeze-dry the raw skins after mounting them on artificial mannikins. In all cases glass, eyes are used to replace the originals. Many taxidermists also use their freeze-dryers to help dry the fleshy parts of specimens mounted by traditional taxidermy techniques.

The quality of the finished product will still depend on the knowledge and skill of the preparator: freeze-drying has never improved the appearance of a specimen.

Although freeze-drying must be regarded as one of the least invasive preparation techniques, freeze-dried specimens may be more liable to insect attack than traditionally prepared items. Insect-proofing chemicals such as Eulan (Edulan U) and Mittel F have been used with freeze-dried material but evidence of their long-term effectiveness is inconclusive (see also Proofing chemicals to prevent attack by insects, p. 4).

Another disadvantage with freeze-drying is that much of the fat within the specimen does not freeze-dry. Hower (1979) maintains that this is not a problem for exhibition specimens if the fat is left undisturbed. Some freeze-drying techniques, however, involve piercing the specimen to speed up drying times. In such cases fat can migrate to the surface of the skin where it causes unsightly staining, before oxidizing to produce a damaging condition known as fat burn.

Furthermore the skeletons of entire freeze-dried specimens are preserved inside the specimen and therefore not available for study. Florian (1990) has indicated that freeze-drying may enhance the deterioration of tissue and that such deterioration may continue during storage. For all these reasons, there has been little interest and much caution regarding the use of freeze-drying for reference collections.

Cole (pers. comm., 1996) maintains that there are very few museums making duplicate collections of freeze-dried material, although the idea of rehydrating chemically unpreserved material in the future seems an interesting option.

Storage of mounted vertebrates

Ramer (1989) has pointed out that mounted vertebrates are more likely to be damaged by poor storage and handling techniques than any other agent of deterioration. This reflects both the low status of such collections in the past and the considerable expense involved in their safe storage. Top of the list of poor storage techniques must be the ubiquitous polythene bag forced over mounted specimens on open shelving.

Small and medium sized mounted vertebrates

Small and medium sized vertebrate mounts used for teaching and reference are best stored in cabinets. If space is available it is good practice to make the base either square or rectangular and keep the mounted specimen within the limits of the base. This will help protect it when being handled, transported and stored.

Steel (1970) describes a system for the storage of mounted birds (although it can also be adapted for mammals) similar to picture storage, where specimens are attached to sliding racks inside purpose-built cabinets. A modified version of this method is to store mounted specimens on both sides of static upright wooden panels within a polythene tent (Fig. 1.16). It provides an economical temporary solution, useful in an emergency.

Large mounted vertebrates

Large mounts are best stored in large spaces. One large space is a far safer and more economical option than several smaller ones. Glass showcases can be adapted and reused for storage, but are difficult to keep air-tight. There may be no alternative to storing large mammals in areas where they are susceptible to dust pollution. Covering them with polythene sheeting is the cheapest in but most depressing solution as it soon accumulates dust electrostatically. The use of magnets to help-seal the polythene as suggested by Guynes (1992) makes access relatively simple.

A preferable but more expensive alternative is to use custom-made Tyvek covers (see Fig 1.8) which can be kept acceptably clean with a clamp cloth. They can be made by sewing, sealing with an impulse sealer, or stapling together. White or unbleached cotton or cotton polyester blends may also be used for this purpose, but make sure, as previously suggested (anon., 1992h), that harmful finishes are washed out.

Game heads may be stored on wall-mounted expanded metal or wire-meshed racks, and covered as described above. Swivel-spring billet hooks are a convenient way of attaching the wooden shields to the mesh (Fig. 1.17). Large horned or antlered heads may be given supplementary support with elastic bungees clipped to the wire. Horn has a tendency to split and flake under clamp conditions, where Morgan (1991) reports that it is particularly susceptible to attack by weevils. He maintains that the RH of areas with horned heads should not exceed 60% (see also Chapter 7 on the Collection environment).

Glass domes and cases

Glass domes and all glass 'Ward style' cases may be simply stored on open shelves with loose dust covers bearing 'Fragile Glass' labels.

Glass-fronted cases should be covered or simply turned to face the wall if stored in areas where daylight cannot be eliminated. With large collections, photographs of the contents may be attached to the backs of turned cases for easy reference.

Frost (1981, 1987) emphasizes that trade labels are part of the history of taxidermy and should be treated with the same care as other natural sciences documentation. If cases with trade labels are to be exhibited, they should be protected from light with hinged acid-free card. If showing signs of damage, labels

Figure 1.16 Keyhole plates used to support mounted specimens on display. Here they are used for upright storage.

Figure 1.17 Example of dog-lead clips (swivel-spring billet hooks) which allow easy access to game heads. 'Bungees' can provide additional support.

Figure 1.18 An original taxidermy trade label which was in danger of deteriorating, now stored in a Mylar envelope

should be removed completely, de-acidified (in consultation with a paper conservator) stored separately in Mylar envelopes (Fig. 1.18) and cross-referenced with photographic records of the case. Van der Reyden (1995) describes techniques for storing archival documentation and, Kishinami (1992) suggests methods for label storage and repair.

Handling, packing, and transportation of mounted vertebrates

Cased material and loan collections housed in their own purpose-built containers pose no particular handling problems. Glass cases should be taped, marked fragile and moved either strapped and palleted, or, if too small or fragile for this, handled with gloves.

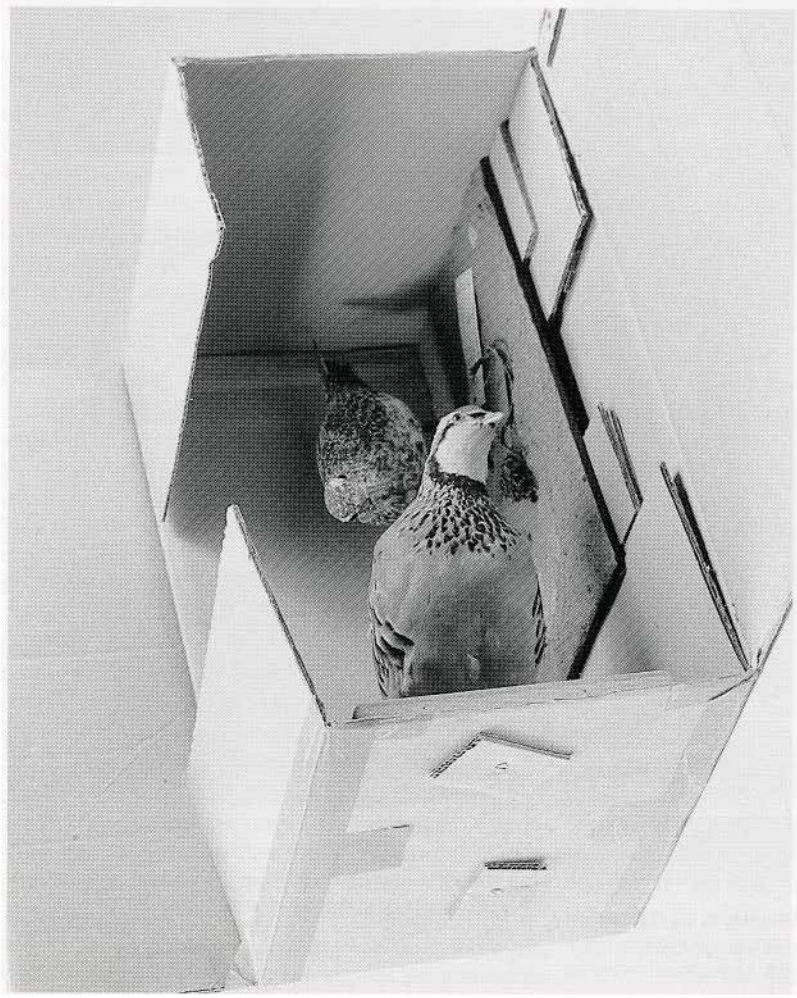


Figure 1.19 An example of more than one specimen securely fixed in the same box. Always indicate the packing method on the outside of the container.

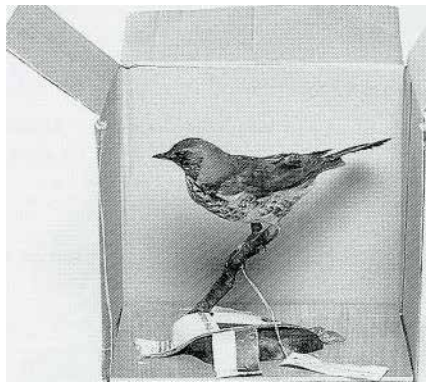
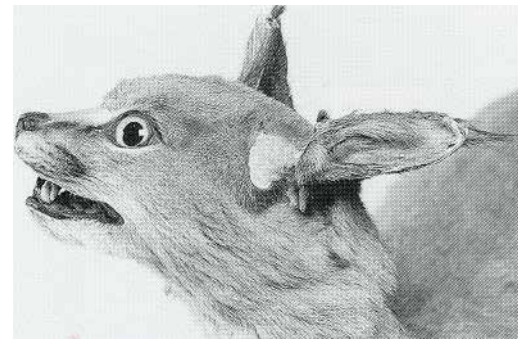


Figure 1.20 Specimens on their original bases should be secured to their travelling container with straps stapeled over the bases.

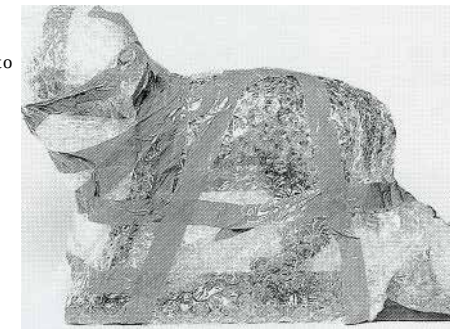
Uncased specimens, however, are at their most vulnerable during any handling procedure. All such material should be mounted on, and only handled by, a substantial base. The most easily damaged parts of mounted birds are their necks. The slightest knock to the bill (b) when moving specimens in and out of cupboards, for example, can cause irreparable damage. The claws of birds of prey are razor sharp, and should be carefully wrapped if items are moved without a supporting base.

Although the tails of mounted mammals will contain an internal wire support, and the ears either a modelling composition around a wire or an artificial ear liner, they are nevertheless the most vulnerable parts of such specimens.

For travel, small and medium sized birds and mammals may be enclosed in a wooden or stout cardboard box. The base should be attached to the box with screws from the outside (Fig. 1.19) or, if documentation, trade labels or the original base are likely to be damaged, strapped and stapled to the bottom (Fig. 1.20). Whatever method is used, it is important to avoid further packing around the animal unless absolutely necessary. More damage has been inflicted on mounted specimens by well-intentioned supporting packing, especially around the heads and tails, than anything else (Figs 1.21a, b and c). If extra packing is essential, a combination of tissue- paper wads, as illustrated in Pye (1992), and polyethylene foam will usually suffice.



(a)



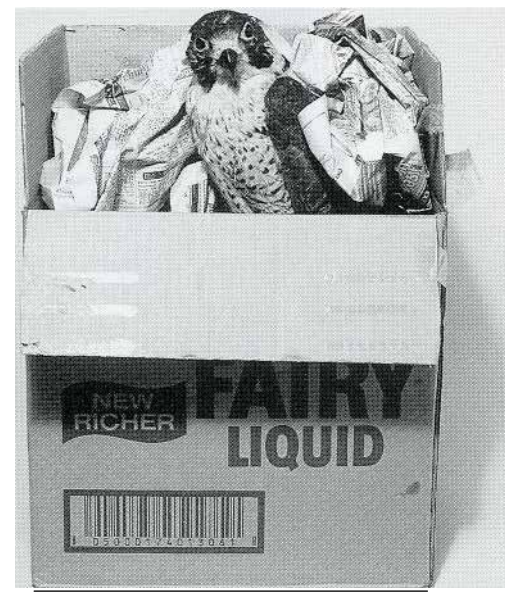


Figure 1.21 Inappropriate packing around specimens can lead to damage during transit (a and b) or whilst unpacking – additional supportive packing such as the ubiquitous crumpled newspaper (c) is usually not necessary if the specimen is firmly attached to its base

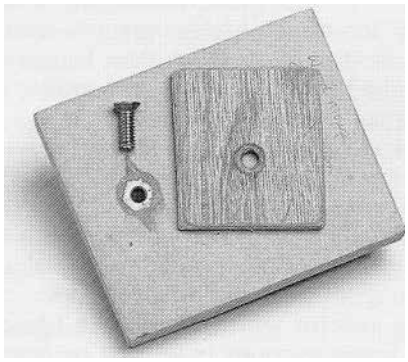


Figure 1.22 The underside of a specimen base showing a recessed nut glued into the bottom. The base is secured to its travelling box with a flat-headed bolt and wooden washer. This simple repacking solution will help reduce damage to specimens regularly on loan.

Packing of returned loan specimens is often carried out by inexperienced people, and should be made as foolproof as possible. Techniques such as gluing a recessed not into the bottom of the base and securing it with a flat-headed countersunk bolt and washer will help define the packing method (Fig. 1.22). Instructions should be clearly indicated on the outside of the box, in the appropriate language, together with a description of the contents.

Large collections of uncased mounted specimens on bases may be safely moved by securing them to flat wooden boards with stapled paper straps. The hoards (the larger they are, the more stable they will be), will support themselves during transportation, although their size will be limited by access constraints.

If large mammals are regularly on loan, they may require their own purpose-built mobile container (Fig. 1.23). Horns or antlers are a particular hazard and should be sheathed in protective material such as polyethylene foam.

The fragile nature of the skins of mounted reptiles, amphibia and fish makes them particularly susceptible to handling damage. Apart from the larger reptiles, this group is often represented by replicas in museum collections. Mounted skins may be the exception, and should be valued as such when assessing storage priorities.

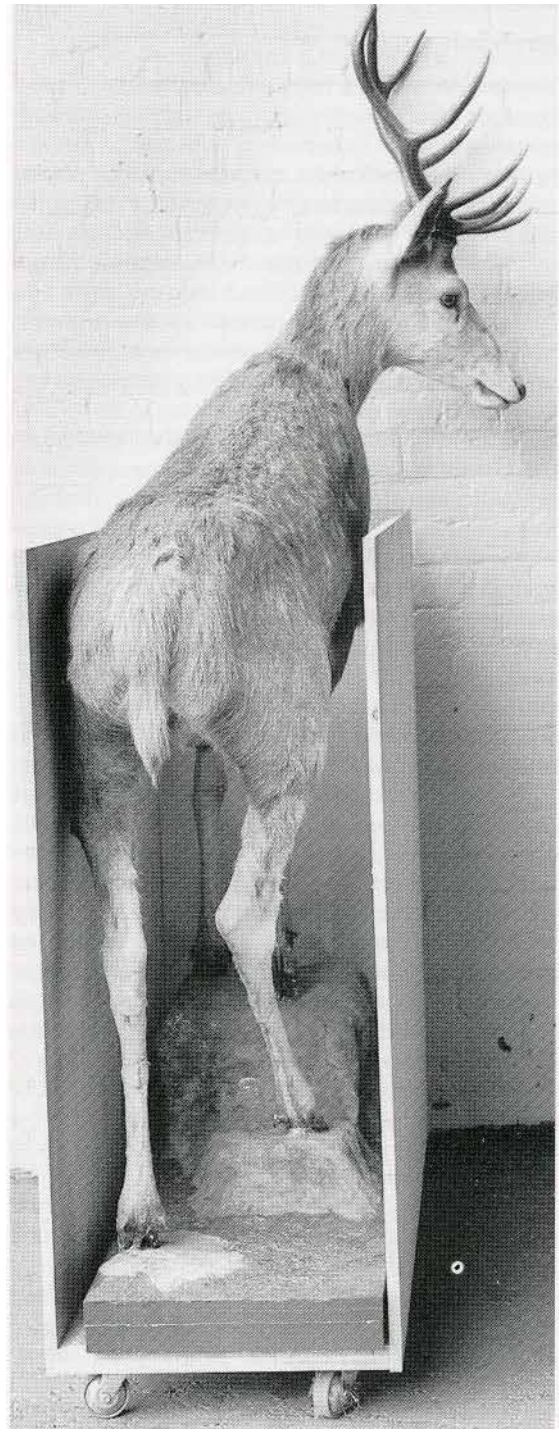


Figure 1.23 A purpose-built mobile container which will ease the strain placed on the legs of some mounted specimens during transportation.

Conservation

Research

The past ten years has seen an increase in the number of research papers in natural sciences conservation. Duckworth *et al.* (1993), however, includes an extensive list of topics awaiting investigation. At present it is still difficult to make informed conservation decisions, let alone confident ones. Although we know which materials and techniques work in practice, we are still uncertain about long-term costs. If in doubt, do nothing is a laudable approach but does not reflect the possibility that no action may mean no collection. The work of Williams (1991) and Young (1992) on the shrinkage temperature of collagen fibre as an indicator of skin deterioration is a promising avenue of research.

There are also ethical questions to be considered. Should we improve upon the poor workmanship of the past, by remodelling mounted specimens to look more realistic, for example? Such decisions should not be made purely on conservation arguments plucked from the fine and decorative arts. Natural science specimens in museum displays often fulfil a different function to those in other disciplines. Sensible decisions on treatments should reflect this.

Whatever decisions are taken, it is imperative that methods and materials are fully recorded either on the specimen card or database, and cross-referenced. This evidence, so lacking in the past, is vital if we are to determine the effectiveness of today's methodologies.

Cleaning

Birds and mammals

Horie (1988) discussed the ethics and science of cleaning vertebrate material and offered some practical suggestions.

The traditional method of cleaning dirty specimens is similar to that adopted for the initial cleaning during preparation (see, Preparation of bird skins, p. 5). Garner (1988) suggests the progressive use of compressed air, mild detergents. organic solvents and dry powders to clean both fur and feathers. The investigations of Rogers (1990a. b) conclude that there is no entirely safe method for cleaning old, dirty bird feathers, although ultrasonics used for cleaning ethnographic feathered items is worth investigating. Bent feathers may he straightened with steam, and broken ones repaired with bamboo splints.

Trodd (1992) recommends that very dirty large mammals and game heads may be cleaned with a mild shampoo of pure soap-flakes. The fur must be rinsed with clean water which is immediately removed using an Aquavac-type vacuum cleaner. Some carpet cleaning systems which combine a fine spray with a water vacuum may also be used on dirty specimens that are otherwise in good condition. Aerosol foam upholstery cleaners have been used successfully, but these leave a powder on drying which must be removed by gentle vacuuming. With all of these techniques the skin must be kept as dry as possible during cleaning.

The Canadian Conservation Institute (anon., 1983) suggests brushing fur (excluding deer) with a mixture of shellac and methyl hydrate before drying and combing out. Perhaps less invasive is the suggestion of Fenn (pers. comm.. 1993), of warm cornmeal or bread-crumbs made from Italian bread containing olive oil for cleaning fur skins. Horie (1988) opposed the use of powdered eraser, a material often used for ethnographical fur skin collections.

In study skin collections, fat burn caused by inadequate initial removal of fat is a prime cause of deterioration. Successful degreasing has been achieved by completely immersing specimens in organic solvents such as trichloroethylene. Robbins (1989) prefers using perchloroethylene, whereas Koch (1991) reports successfully degreasing bird skins with a variety of fur-soaking chemicals. All such work is hazardous for the preparator and should only be carried out whilst wearing the correct personal protective equipment and in areas with adequate ventilation. The success of these methods will often depend on the thoroughness of the initial preparation and the type of skin being degreased (see also Preparation of bird skins, p. 5). Injecting friable skins with rubber latex, a traditional solution for fragmenting skins, is not to be recommended for their long-term conservation. Latex is difficult to reverse safely and will itself deteriorate.

Reptiles, amphibia and fish

In the past, many reptiles, amphibia and fish were coated with shellac to seal them and make cleaning easier. Shellac discolours badly with age and can be extremely difficult to remove. As a very last resort Entwistle (1992) used a paint remover containing 4.5% sodium hydroxide. For practical purposes the technique was very successful, but the long-term effects have yet to be assessed.

Osteological material

Adams (pers. comm., 1982) suggests that an ultrasonic bath containing 2% non-ionic detergent may be used for small skeletons. It requires about thirty seconds for small-mammal sized specimens.

Larger material may be cleaned with the same solutions but this requires the pressure of a paint spray-gun to help remove ingrained dirt. High pressure jets should be reserved for worst-case exhibition material only. All specimens should be thoroughly rinsed with water after cleaning.

Large skeletons can be successfully cleaned *iii sins* with controlled cryogenesis (spraying with dry ice), (anon., 1996). This technique is very much in its infancy but may, in the future, have some application in the natural science field.

Restoration and consolidation

There has been little published work on the restoration of dry vertebrates. The papers by Rau (1993) and Hildebrand (1985) on the remounting of Quaggas at Mainz, Munich and the South African Museum are a unique source of reference. The work was experimental and time will judge its success.

The traditional method of relaxing the split seams of mounted vertebrates with water and restitching them can be successful, at least in the short term. Horie (1988) advises caution with this technique, as wetting the skin can result in increased stiffness on drying and compound the problem. Howard (1989) does not attempt restitching, preferring to cut away the loose edges of split skin before filling and retouching.

Cracks or splits in the skins of fish, reptiles and amphibia are very common. Entwistle (1992) reports that successful repairs have been achieved with fibreglass tissue and polyvinyl acetate emulsion.

In 1988 the Canadian Conservation Institute began a series of trials with Paralene, a modern polymer, to consolidate natural science materials and make them easier to handle and clean. The advantages of coating fur and feathers were minimal but, as Grattan and Morris (1991) point out, it did help consolidate friable bone, strengthen bird eggs and improve the handling possibilities of some reptile skins. However, its use may be limited in the conservation field by the irreversible nature of the process.

Relaxing mounted specimens

Mounted rarities, mounted specimens with scientific data and large flying birds may be relaxed and prepared as cabinet skins in order to preserve them, or simply to save space. This can be a damaging procedure, as the ability of a skin to relax depends largely on its initial preparation, and serious consideration should be given to the consequences of such decisions. Wagstaffe and Fidler (1968), Summers (1979) and Hangay and Dingley (1985) describe techniques for relaxing skins and should be consulted before proceeding.

Many of the leading natural history museums around the world recognize the popularity of mounted specimens, and have used information technology and interactivity to enhance the appeal of their displays. It would be a mistake to confuse modernising displays with the elimination of mounted vertebrates. Replacing them with two dimensional images only duplicates what can be found in other venues and ignores the interest engendered by real specimens, the heart of the museum collections.

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Suppliers

Leather Conservation Publications. The Leather Conservation Centre, 34 Guildhall Road, Northampton NHI 1EW, UK.

Squirrel Data Logger, Grant Instruments, Barrington, Cambridge CB2 5QZ, UK.

Taxidermy Supplies, Europe Eyes, 29 Greenford Close, Orrell, Wigan, Lancashire WN5 8RH. UK.

Lorne Taxidermy Supplies, Kilnhillock Cottages by Cullen, Buckle, Banffshire AB56 2TB, UK.

Snowdonia Taxidermy Supplies, Llanrwst, Gwynedd, N. Wales LL26 OHU. UK.

The Guild of Taxidermists

The Guild of Taxidermists was established in 1976 to raise and maintain the status and standards of taxidermy in the UK. Membership is open to all interested in the subject and is not, in itself, a qualification. The Guild does offer its own qualifications based on quality of work and judged by a panel of experienced taxidermists. The basic qualification. Professional Membership, acknowledges a good standard of work across the whole discipline. The Guild is a source of advice on all aspects of taxidermy and, for advice on commissions, will recommend the names of Professional Members.

Formulae

Huber's Fluid

Petrol	10 litres
I.M.S.	630 ml
Turpentine	130 ml

Rowley's mixture for osteological work

Pancreatin	80g
Sodium sulphide	40g
Water	5 litres