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ARCHIVAL STORAGE OF DISINTEGRATING LABELS FROM FLUID-PRESERVED SPECIMENS

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Abstract.—The disintegration of paper labels attached to alcohol-preserved specimens is a common curatorial problem. A method of removing, replacing, repairing, and storing these labels before the data are irretrievable has been devised. A storage system for 35mm film negatives, with mylar and acid-free paper components, has been adapted for this purpose. This filing system provides an archival-quality storage environment that prolongs the life of the labels and allows for convenient retrieval. Handling protocols ensure that both replacement labels and catalog cards clearly state that the original label was removed, when it was removed and by whom, and where it is to be found.

A common problem in all fluid-preserved collections is the eventual disintegration of paper labels. The Vertebrate Zoology Collection of the Bishop Museum has been experiencing such disintegration, especially in labels from specimens dating from the early 1900s through the 1960s. In some cases, even labels on specimens less than a year old have shown signs of deterioration.

Hawks and Williams (1986) stated categorically that labels should never be discarded. Indeed, the importance of preserving specimen labels, especially the original collector's label, cannot be overstated. The original collector's label may be the primary source of collection data, and in the absence of a collector's catalog, may often be the only source of collection data. When transcribing data to a new label, errors may be made, even by the most careful of workers. If the original label has been discarded, there may be no possibility of correcting a mistake. Another important reason to keep specimen labels is to compare writing styles. Illegible writing or ambiguous data can often be deciphered when compared to another label or series of labels in the same hand. Recognition of the necessity of preserving original specimen labels, and a growing awareness of the serious degradation of many of the labels attached to the fluid-preserved specimens in our collections, led to the development of our current procedure to preserve disintegrating labels.

STORAGE SYSTEM

Before 1983, our practice was to fold or roll completely or partially torn labels and resecure them to the specimen. This was not satisfactory, because the paper continued to deteriorate. In 1983, we investigated several storage arrangements, including storing the original labels with the accession file, with the catalog, or in a separate "original label" file. Storage in the accession file was rejected because relocating the labels would require too many steps. Storing the labels with the catalog was also unacceptable because our catalog is a card file, which would require the label to be glued, taped, stapled or clipped to the catalog card.

The third option was to create a separate "original label" file. The specific attributes required were: 1) an environment that promoted the longevity of the labels; 2) easy cross-referencing of label to specimen; and 3) efficient retrieval of labels. The paper conservators of the Pacific Regional Conservation Center (PRCC), based at Bishop Museum, suggested that we use the Light Impressions Company's Nega*Guard® System, developed for film negative storage. It consists of mylar sleeves designed to hold 35mm film negative strips, acid-free folders designed to hold several of the mylar sleeves, and an acid-free box to hold the folders (suppliers listed in Appendix).

Before the original label is removed from a specimen a replacement label with all the data is written. In addition, "Original label disintegrating, removed to old label file," the date of removal and the name of the person performing the task is added to the new label. (See Hawks and Williams (1986) and Williams and Hawks (1986) for recommendations concerning paper, thread and ink.) The original label is then removed and placed in a distilled water rinse, and the replacement label is attached to the specimen. Rinsing the label is especially important when the alcohol in which the specimen and label have been stored has become discolored with body fluids and suspended pigments and oils, and there are bits of feathers, fur, or scales that may cling to the label. The rinse water is changed until it remains clear (a 100 ml beaker is useful for rinsing individual labels). The final rinse is done with distilled water to which has been added a saturated solution of calcium hydroxide to bring the pH to between 7 and 8. This solution must be prepared just prior to use and the pH should be verified with a pH meter or pH strips (see Clapp, 1987:85–86, 146 for a discussion of the use of and recipe for mixing calcium hydroxide solution). After soaking in the final rinse for approximately 10 minutes, the label is placed on an acid-free blotter and gently dried with a second blotter. When it is no longer wet, but merely damp, it is sandwiched between 2 layers of Reemay, a thin, non-woven polyester fabric, and this in turn is sandwiched between 2 layers of acid-free blotter. The whole stack is then weighted under plate glass and supported on a smooth, flat surface such as a formica desk top. The polyester fabric between label and blotter paper prevents the two from sticking to each other as the label dries. The blotters are changed 2 or 3 times until the label is dry. When the label fragments are dry, the catalog number is written on each piece that does not already bear the full number. Only carbon ink or lead pencil are used.

Completely or partially torn labels are common and must be repaired before storage to prevent loss of individual pieces. Hawks and Williams (1986) found two repair methods particularly useful in mending specimen labels: the starch paste and Japanese tissue method described by Clapp (1987:105) and Ritzenthaler (1983:102), and the heat-set tissue method described by Roberts and Etherington (1982:130). These methods were found to be acceptable for labels in dry storage and should prove equally effective for dried labels removed from wet storage. In this case, however, the PRCC paper conservators recommended the use of Archival Aids Document Repair Tape. This is a very thin, transparent, acid-free, adhesive tape that is advertised as being completely reversible. As its reversibility had not been tested by the conservators, they recommended that we apply it only on one side of the label and only over the actual juncture of the two pieces being joined, not over the whole label. The tape must be burnished firmly to ensure good contact between tape and paper. The tape does not provide any strength to the label; it merely holds the pieces together. The mylar sleeve in which the label is stored provides the necessary support to labels weakened by tears.

Once the necessary repairs are made to the label it is placed in a clear mylar

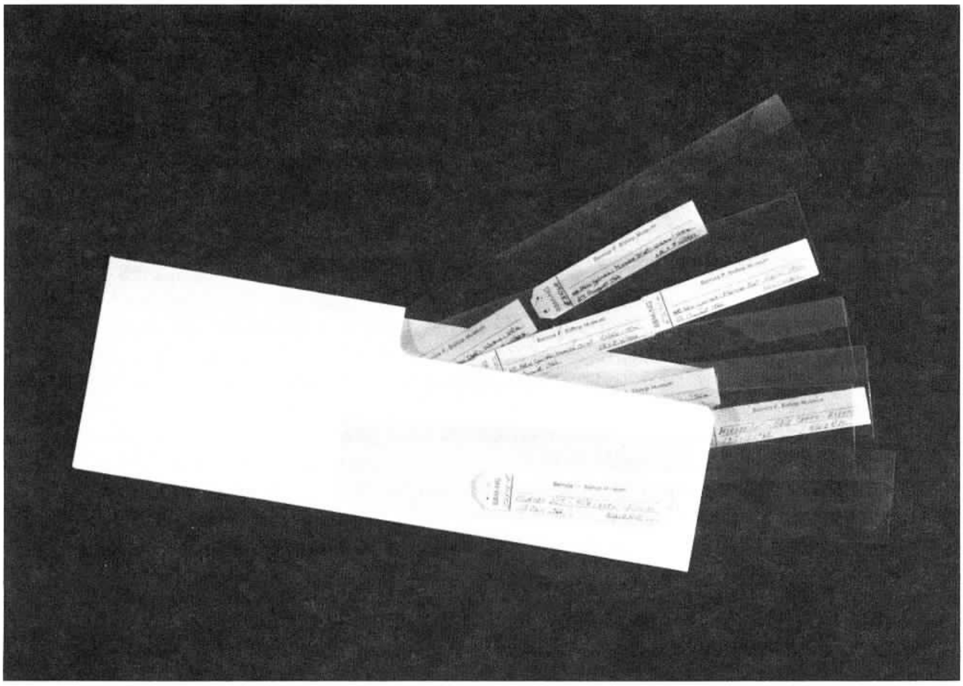

Figure 1. Several mylar sleeves, each containing 2 to 3 original labels, are stored in one acid-free folder.

sleeve which allows a user to read both sides without handling the label. Depending on size, two or three labels can usually be placed in one sleeve (Fig. 1). Several mylar sleeves can be grouped into each acid-free folder. On the outside of the folders the catalog numbers are written very lightly, in pencil, so that adjustments are easily made when new labels are added. The folders are stored in an acid-free box that is part of the Nega*Guard® System.

We pre-assign catalog number series to the file folders in groups of 250 or 500 to eliminate the need to shuffle labels and mylar sleeves as the number of stored labels increases. The number series assigned to each folder is based on a guess as to how many specimens in a number series are preserved in alcohol, and how many of those may actually need to have original labels replaced.

Finally, the catalog card is annotated with the removal statement that was written on the replacement label. This serves as backup and may facilitate researchers who wish to see the original label. To ensure that the removal statement is not forgotten, attached to the inside of the file box lid is a sample catalog card and label showing the preferred placement and wording.

Conclusions

Almost 5 years of use has shown that the label storage system described above is a convenient and effective method of preserving and organizing disintegrating labels of fluid-preserved specimens. The removed label is immediately replaced with a good quality substitute label, rinsed, dried, repaired (when necessary) with an archival-quality, acid-free tape, and stored in a non-reactive, acid-free environment. Both replacement label and catalog card are annotated with a standard removal statement. And lastly, the labels are filed numerically in a separate file and are efficiently retrieved.

Acknowledgments

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Appendix: Suppliers

Acid-free, lignin-free file box; acid-free, lignin-free folders; and Mylar D negative storage sleeves.

Sold as Nega*Guard® System from Light Impressions, 439 Monroe Ave., Rochester, NY 14603-3717.

Sold as Archival Lig-Free® II Photo-System from Conservation Resources International, 8000H Forbes Place, Springfield, VA 22151.

Archival Aids Document Repair Tape.

University Products, Inc., P.O. Box 101, 517 Main St., Holyoke, MA 01041.

Conservation Materials Ltd., 240 Freeport Blvd., Box 2884, Sparks, NV 89431.

Blotter paper.

Archivart Lightweight and Heavyweight Blotting Paper, Process Materials Corporation, 301 Veterans Boulevard, Rutherford, NJ 07070.

Conservation Quality Blotting Paper, University Products, Inc., P.O. Box 101, 517 Main St., Holyoke, MA 01041.

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TALAS, 213 West 35th St., New York, NY 1001-1996.

HEALTH CONSIDERATIONS OF RADON SOURCE FOSSIL VERTEBRATE SPECIMENS

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Abstract.—Vetebrate fossils from the Jurassic Morrison Formation, Plio-Pleistocene Hagerman Lake Beds, and Oligocene Cypress Hills Formation of western North America, and the Permian Karoo Sequence of South Africa have been found to be radioactive and are radon sources. Radon is an odorless, tasteless, and invisible radioactive gas formed during the decay of uranium. Fossil vertebrates may concentrate the radon precursor, radium, because of its chemical similarities to calcium. Radon daughter elements collect on airborne dust and, if inhaled or ingested, increase the risk of cancer. Good ventilation and prudent handling techniques are recommended in areas where radioactive specimens are present.

VERTEBRATE FOSSILS CONTAINING RADON

The Upper Jurassic Morrison Formation has a vertebrate biota consisting predominantly of reptiles, with a few small mammals. Dinosaurs from Colorado, Utah, and Wyoming have been collected, stored, and displayed in museums for over a century. First described by Cope (1877) and Marsh (1877), this formation has yielded specimens of *Apatosaurus, Brachiosaurus, Stegosaurus, Allosaurus, Diplodocus, Camarasaurus, Dryosaurus,* and other species.

The Upper Pliocene-Pleistocene Hagerman Lake Beds of southern Idaho contain a vertebrate fauna that has been dated at 3.4 million years (potassium-argon dating). Discovered in 1928 near Hagerman, Idaho, the site is rich in zebra-like horses, muskrats, beavers, otters, fish, frogs, cormorants, geese, swans, ducks, pelicans, and other animals (White, 1967). This abundant fauna has been studied, made available for study, or put on public display by a number of museums.

The Lower Oligocene Cypress Hills Formation of southwestern Saskatchewan, Canada, contains a rich mammalian fauna of small and large species including *Brontotherium, Titanotherium, Archaeotherium, Stylemys,* and *Anthracotherium.* Specimens collected from a fluvial conglomerate of the Cypress Hills Formation in the Hunter Quarry near Calf Creek and from other localities are maintained at the Royal Ontario Museum (ROM) and the Saskatchewan Museum of Natural History (SMNH). These specimens were found to be radioactive by ROM, SMNH, and the National Museums of Canada.

The Permian Karoo Sequence of South Africa has a rich vertebrate fauna that emits low level radioactivity (van den Heever, personal communication). Karoo reptiles include *Eunotosaurus, Tapinocephalus, Endothiodon, Cisticephalus,* and *Pareiasaurus* (Romer, 1966). Because radioactive elements occur naturally in some rock and soil deposits, other fossil vertebrate faunas may be radioactively hot.

Radon occurs naturally not only in soil and water, but also in rocks such as granites, shales, and phosphates containing radioactive minerals such as carnotite, uraninite, and pitchblende. Radon also occurs in soils contaminated with industrial wastes such as the byproducts of uranium or phosphate mining and pro-

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cessing. Chemical similarities between the radon precursor, radium, and the calcium component of bone may cause radium to be concentrated in nearby vertebrate fossils (Stine, 1979). Associated uranium minerals such as uraninite and carnotite are the radon sources in the fossil-bearing Morrison and Karoo strata. The radon sources for the Hagerman and Cypress Hills faunas are unknown, although uranium is mined at several localities in Idaho.

Health Hazard Awareness

The radon health hazard is associated with the products of radon decay, called radon daughters. These daughter products are solids that are attracted to dust motes which are then inhaled and get trapped in the lungs. The only known health effect associated with long term exposure to elevated levels of radon is an increased risk of pulmonary cancer. Certainly not everyone exposed to elevated levels of radon will develop cancer, and the time between exposure and onset of the disease may be many years. The risk is cumulative and is dependent on concentration of radon and length of exposure. The U.S. Environmental Protection Agency (U.S. EPA) estimates that radon is responsible for approximately 5,000 to 20,000 deaths annually in the United States. Current radon risk information is based on studies of miners who are exposed to varying levels of radon in underground mines (Sax, 1979). There are problems extrapolating these data to average public or workplace environments.

Concern about indoor concentrations of radon began in the early 1960s when homes and buildings built with materials from uranium mine waste were found to contain high radon levels. Recently, significant radon levels have been found, particularly in basements, as a result of infiltration from groundwater and soil vapours. Domestic accumulations can pose substantial health risks in particular geologic areas. The risk of lung cancer is significantly increased when radon exposure is combined with smoking.

Twenty years ago the American Museum of Natural History (AMNH) in New York registered radioactivity in Morrison Formation and Karoo Sequence fossils. The AMNH subsequently isolated their radioactive paleontologic specimens in a separate room. Except for AMNH, no other museums that were contacted have implemented special storage or treatment of their Morrison, Hagerman, Cypress Hills or Karoo specimens. These museums include: Denver Museum of Natural History, Field Museum of Natural History, Idaho Museum of Natural History, Los Angeles County Museum of Natural History, Peabody Museum of Natural History, Royal Ontario Museum, Saskatchewan Museum of Natural History, South African Museum, Tyrrell Museum of Palaeontology, United States Natural History Museum-Smithsonian Institution, and University of Michigan Museum of Paleontology.

Radon Monitors

The U.S. Environmental Protection Agency (1987a) recommends radon checks and radon gas monitoring systems to help reduce the risk of exposure. Problems associated with attempts to measure indoor radon and radon decay product concentrations may arise because of 1) nonstandardized procedures; 2) different conditions prior to and during measurement; 3) seasonal and other weather conditions; and 4) different interpretations of the results. Stabilized conditions, with

Figure 1. Morrison Formation's *Apatosaurus excelsus* in public display area, Dinosaur Hall, Field Museum. Note radon detector mounted on steel framework.

no ventilation, air conditioning, or heating present, produce the least variable measurements.

The two most common monitors, activated charcoal adsorption with a test period of three to seven days duration, and alpha track detectors requiring a two to four week test period, are commercially available to measure radon concentrations. Other techniques with relatively short measurement periods include: continuous radon monitoring, radon progeny integrating sampling (flow-rate air pump), and grab sampling (air drawn through a filter) (Environmental Protection Agency, 1987b). Measurements are reported as "Working Levels" (WL) and as picocuries per litre (pCi/l), and tell more about radiation levels than about radon or radon daughters.

Alpha track detectors were placed in a specimen cabinet, in a storeroom, and in a public display area at the Field Museum for three months, from March 10 to June 10, 1988, to measure the average radon emission of Morrison dinosaurs.

Figure 2. Oversize storage room at Field Museum with Morrison Formation *Brachiosaurus altithorax* vertebrae in foreground. Note radon detector mounted on stairwell brace.

The results indicate that the public display area (Fig. 1) contained 0.9 pCi/l, and the storage room (Fig. 2) contained 3.6 pCi/l. These values are within the range of average background radon concentrations and reduction of the levels would be difficult to accomplish. The storage cabinet housing the radioactive specimens (Fig. 3) indicated a level of 53.0 pCi/l, substantially above the background level. The opening of cabinets containing radioactive specimens may thus present a serious exposure risk.

The U.S. EPA provides free literature on radon and radon testing (U.S. EPA Regional Office, 230 S. Dearborn St., Chicago, IL 60604). Some state or local government agencies provide detectors or radon testing services.

Action

Collectors, researchers, museum personnel, and visitors who come into contact with radon contaminated fossils should be aware of the risk of exposure to airborne

Figure 3. Specimen cabinet in fossil reptile collection area that houses Morrison Formation *Allosaurus* femur. Note radon detector mounted in cabinet door.

radioactive microscopic dust. Diluted with large volumes of air, radon poses little danger. In areas where radon has been detected, ventilation from outdoor air (not recirculated indoor air or a heat recovery ventilator) should be used to increase air flow. Heating and air conditioning costs will increase, but only the exchange of air will satisfactorily dilute the radon concentration. Air cleaners have been found to be ineffective. In addition, the air should be as dust free as possible. Without dust, radon daughters cling to walls and other surfaces where they are little or no hazard.

Known radon emitting specimens such as the Morrison, Hagerman, Cypress Hills, and Karoo material, should be kept in well ventilated cabinets and rooms. Workers handling radon source specimens are urged to use appropriate respiratory and dermal protection, such as particulate filter masks and disposable latex gloves, and to wash hands after handling specimens. Activated charcoal in muslin or cheesecloth bags, placed in storage cabinets, will absorb some emissions.

Although not well documented, radioactivity in vertebrate fossils does constitute a health safety concern in some museums. A survey should be undertaken to determine whether radon hazards are present, and appropriate steps be taken to alleviate them.

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EFFECTS OF INITIAL PREPARATION METHODS ON DERMESTID CLEANING OF OSTEOLOGICAL MATERIAL

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Abstract.—The influence of different processing procedures on the osteological cleaning abilities of dermestid beetles was tested using dried skulls originating from fresh, frozen, and fluid-preserved specimens. The objective was to identify preparation procedures that can provide optimal conditions for effective cleaning of skeletal material. Standard drying of fresh and frozen specimens proved to be the most suitable for dermestid cleaning. Although there are problems associated with some fluid-preserved specimens there are preservation benefits that might encourage its use.

Among the many described methods of preparing vertebrate skeletons, the use of dermestids (Coleoptera: Dermestidae) has been found to be the most efficient and effective (Borrell, 1938; Case, 1959; Hall and Russell, 1933; Hooper, 1950; Russell, 1947; Sommer and Anderson, 1974; Tiemeier, 1940). It is generally accepted that suitable environmental conditions for dermestids are prerequisite for effective cleaning of skeletons (Peterson, 1964); however, initial preparation methods may also influence the dermestid's ability to remove non-osseous tissues.

Drying skinned, eviscerated specimens reduces the risk of putrefaction and consequent degradation, and prepares them for dermestid cleaning. In situations of high relative humidity, drying efforts are often slow and ineffective, thus encouraging bacterial or fungal growth, particularly at higher temperatures. Such growths can make the material less suitable for dermestid cleaning. The drying of skulls is sometimes aided by removing the brain, which may also reduce cleaning time for dermestids. Lucas (1950) suggests using a probe to remove the brain, whereas some suggest forcing water into the braincase (McComb, n.d.; Schmitt, 1966) and others suggest soaking the skull before forcing water into the braincase (Hall, 1962).

Fluid preservation of specimens eliminates the need for immediate drying. However, skulls removed from specimens that have been treated with formalin are difficult to clean with dermestids (Sommer and Anderson, 1974; de la Torre, 1951). Old specimens and specimens coated with other chemical preservatives are also problematic for dermestid cleaning (Case, 1959; Hall, 1962). Some authors recommend application of fats or oils to stimulate cleaning of such material (Hooper, 1950; de la Torre, 1951). There is a variety of preparation techniques and supplemental treatments that may be used on osteological material before it is placed with dermestids for cleaning.

If it is possible to identify preparation techniques that are most suited for dermestid cleaning, it may be possible to develop these techniques to provide high-quality preparations that require little or no subsequent treatment. With this consideration in mind, we have experimentally determined the effect of various preparation techniques on dermestid cleaning. Emphasis was given to standard methods of initial specimen preparation, including drying, soaking in water before drying, brain removal, ethanol preservation, formalin-ethanol preservation, formalin-ethanol preservation with a subsequent ammonia treatment, and freezing.

Table 1. Summary of preservation treatments used on samples.

Sample	Soaked in water	Brain removed	Frozen	Fixed in formalin	Stored in ethanol	Soaked in ammonia	Dried indoors	Dried outdoors
A							X	
B								X
C	X	X					X	
D	X	X						X
E				X	X		X	
F				X	X	X	X	
G					X		X	
H			X				X	

Methods and Materials

Bat specimens (*Artibeus jamaicensis*) from the Caribbean, collected in May, 1986, were prepared using different treatments as summarized in Table 1. Skulls from Samples A, B, C, and D were removed from fresh specimens; C and D were initially soaked and the brains were removed; B and D were dried outdoors. Samples E and F had skulls removed from specimens stored several months in 70% ethanol (initial treatment involved fixation in 10% formalin for three to four weeks followed by 24 hours of rinsing with running tap water); F was subsequently soaked in 25% ammonia before drying. Skulls from Sample G were removed from fresh specimens and stored 11 months in 95% ethanol. Sample H had skulls removed from specimens that had been frozen for five months. Skulls in all of the samples were cleaned at the same time by dermestids in June-July 1987.

Each dried skull was weighed before and after dermestid cleaning to determine the amount of tissue removed. Weights were measured with a Mettler balance (Model H35AR) which provides readings of 0.0001 gram. Each skull was placed in a jar with 10 larvae (10–15 mm in length) of *Dermestes maculatus* and a small amount of cotton. The jars were covered with screen. Progress of the cleaning was monitored for 60 days. Skulls were removed when they appeared to be thoroughly cleaned. The number of days required for cleaning was recorded for each skull.

Survivorship of larvae feeding on tissues prepared in various ways was documented by recording the numbers and stages of dermestids remaining in each jar when the skull was removed. Additional testing was conducted to confirm the effect of ethanol treated tissues on dermestid survivorship. One hundred adult dermestids were placed in containers with the dried ethanol-treated or untreated tissues for four days to lay eggs and then removed. The eggs were allowed to hatch and larvae were observed as they progressed through succeeding instars. Pupae were removed for metamorphosis so that emerging adults would be isolated and fertility could be assessed. Further comparisons were made to determine dermestid preference for fresh or ethanol-treated tissue. Four samples of muscle tissue, each weighing about 65 grams, were removed from the same animal and treated in different ways. One sample was soaked one week in 70% ethanol; another was soaked one week in 20% ethanol. A third sample was frozen, then removed and dried for two weeks with the first two samples. A fourth sample was frozen, then thawed and dried overnight. All four samples were placed in an aquarium stocked with dermistid larvae. Dermestid preference was determined by daily monitoring of weight loss until all tissue was consumed.

The quality of dermestid cleaning was evaluated by noting with 20× magnification the quantity of non-osseous tissue remaining on the interorbital region. This region could be easily examined and compared. It also represents an area where the non-osseous coverings range from thin connective tissues (on the rostrum) to massive temporalis muscles (on the braincase). The quality of cleaning provided by the dermestids was categorized in the following manner: 1 = virtually no tissue present; 2 = minute traces of tissue present; 3 = small, visible amount of tissue present; 4 = large amount of tissue present (Fig. 1). Also, the amount of dermestid damage to paper specimen tags (fluid-resistant labels from Texas Tech University labeled with Pelikan 17 Black Ink) was ranked as: 1 = no visible loss of tag or associated data; 2 = minor damage with some visible loss of tag parts, but not affecting data; 3 = major damage with obvious loss of tag parts, sometimes including data (Fig. 2).

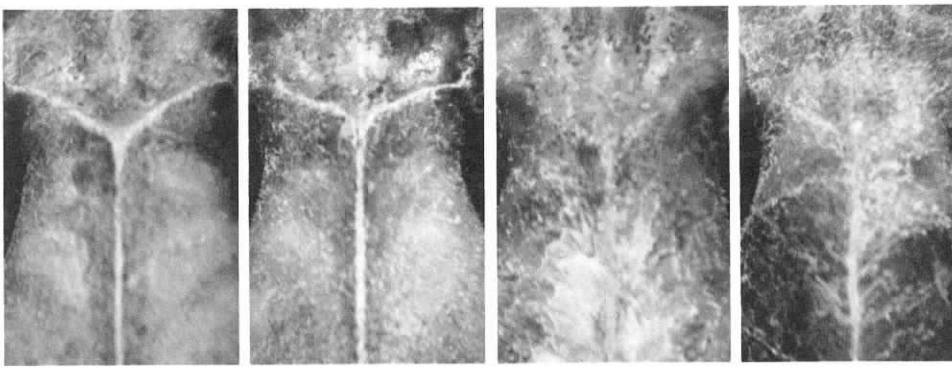

Figure 1. Interorbital regions of skulls showing cleaning quality by dermestids. Photographs are arranged from left to right according to the ranking of 1 to 4 described in the text.

Results

Table 2 summarizes cleaning time and amount of tissue removed. As expected, specimens with brains removed (Samples C and D) were cleaned faster because there was less tissue to be consumed. Skulls initially stored in ethanol (Sample G) also had a rapid cleaning rate even though the brains were intact. The average weight of tissue removed from Sample G skulls was less than averages of other samples that had brains intact (A, B, E, F, and H). It was assumed that these weight differences were primarily attributed to fats being dissolved and removed from the skull by the ethanol. Samples receiving formalin fixation (E and F) required the longest cleaning times; there was a slight improvement in the time and quality of cleaning in Sample F, which was soaked in ammonia before drying.

Some preservation procedures may directly affect the dermestids, particularly procedures involving chemicals. For Samples C, D, and G, the cleaning time was so brief that no deleterious effects were evident, except some pupating individuals may have been cannibalized. For Samples A, B, and H, individuals frequently

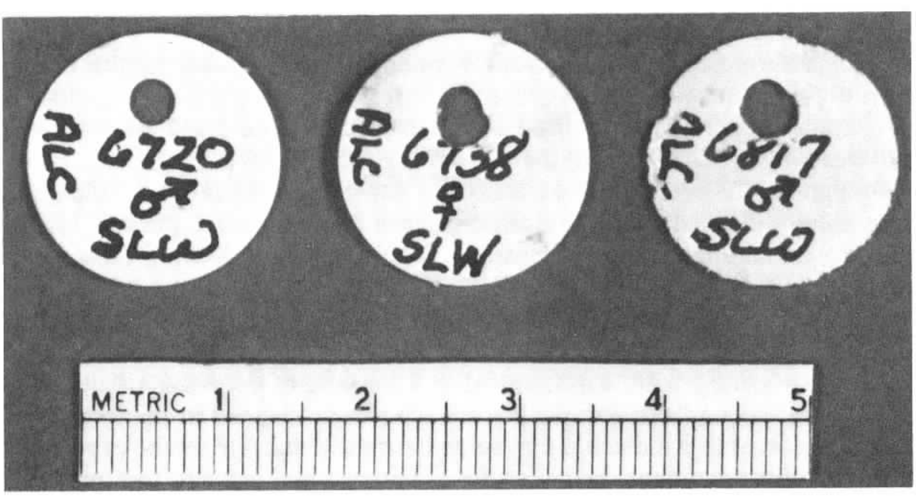

Figure 2. Photographs of specimen tags showing degrees of dermestid damage (LEFT = no damage; CENTER = minor damage; RIGHT = major damage).

Table 2. Summary of dermestid cleaning activities asociated with study. Methods of determining time, quantity of tissue removed, and rankings for quality of cleaning and tag damage are described in text.

Sample	Sample size	Time avg (min–max) (days)	Quantity avg (min–max) (grams)	Quality of cleaning (frequency of ranks) 1	2	3	4	Tag damage (frequency of ranks) 1	2	3
A	10	29.3 (10–53)	0.9 (0.7–1.2)	2	7	1	0	3	6	1
B	10	26.8 (15–46)	0.9 (0.8–1.1)	5	2	3	0	5	5	0
C	10	17.2 (10–30)	0.6 (0.5–0.7)	0	8	2	0	0	8	2
D	10	13.1 (10–21)	0.5 (0.5–0.7)	0	10	0	0	3	5	2
E	10	60+	0.8 (0.6–0.9)	0	0	0	10	0	4	6
F	9	41.4+ (15–60+)	0.8 (0.6–0.9)	1	5	0	3	1	1	7
G	12	17.1 (10–37)	0.7 (0.7–0.9)	1	3	8	0	0	2	10
H	12	25.4 (15–37)	0.9 (0.7–1.1)	1	5	6	0	3	9	0

developed into adults and laid eggs that provided new larvae. Similarly, dermestids feeding on specimens preserved with formalin (E and F) laid eggs but had considerably fewer hatchlings. The survivorship of original individuals in these samples was low, particularly after 30 days. For the 14 specimens that were not cleaned at the end of two months (all of E and part of F), the survivorship of original individuals averaged 6.4%. Obviously, the low numbers affected cleaning ability.

Of the chemically treated samples examined, the cleaning time of Sample G was so brief (average = 17.1 days) that the effect of ethanol-treated tissues on dermestids was not evident, thus further investigation was warranted. It was initially determined that inadequately dried ethanol-treated tissues could be fatal to adult dermestids. Additional testing showed that the hatchling larvae raised solely on dried ethanol-treated tissue had lower survivorship and a longer maturation period than those associated with untreated tissues. After four months of observation only 10 dermestids from the test group reached adult stage compared to 283 in the control group. However, the adult dermestids raised entirely on ethanol-treated tissues were fertile and offspring appeared to develop normally if fed untreated muscle tissue.

Although dermestid larvae eat both fresh and ethanol-treated tissues, there is a definite preference for untreated tissues. Based on feeding sequences, the order of preference was fresh tissue dried overnight, fresh tissue dried for two weeks, tissue treated with 20% ethanol, and tissue treated with 70% ethanol.

Although skulls were removed from the dermestids when they superficially appeared to be cleaned, closer examination with 20× magnification provided the detail for evaluating quality of cleaning. The results of cleaning rankings are summarized in Table 2. Very few skulls were cleaned well enough to be ranked as 1; Sample B had the greatest number with 50%. Most skulls from other samples had minute amounts of tissue remaining (ranked as 2 or 3). Samples E and F were the only groups with skulls ranked as 4 because of excessive tissue present after 60 days; no skulls in Sample E were adequately cleaned in the test period.

Examination of tags indicated that some dermestid damage can be expected for any preparation treatment used. Results of damage rankings are summarized in Table 2. Very few tags showed no damage. Most minor damage (rank 2) was associated with the hole used for string attachment. In these cases, it appears that the larvae widened the hole in order to crawl through. In cases of major damage (rank 3) the dermestids were feeding on the tag, thus such damage represents a primary concern in evaluating preparation procedures. The samples associated with fluid preservations (E, F, and G) showed the highest incidence (=60%) of major tag damage, compared to all other samples (=20%). Because these groups had average cleaning times ranging from 17.1 to 60+ days, no relationship can be made between damage and the amount of time the tags remained with the dermestids. There were no situations where the tag was destroyed beyond legibility; only a couple required data verification because parts of the labeling were missing.

Discussion

The results of this study showed that commonly used air-drying procedures are compatible with dermestid cleaning. There was no benefit for dermestid cleaning obtained with different drying rates resulting from indoor and outdoor drying. If specimens are small enough that the brains will dry quickly, removal of the brains may not be justified, particularly if such tissues can be removed without damage by the dermestids. Furthermore, brain removal usually involves soaking the entire fresh skull in water, thus saturating other tissues and prolonging the drying process.

Our results indicate that chemically-treated tissues may affect the survivorship of dermestids. Although ethanol-treated specimens may be toxic to dermestids, properly dried ethanol-treated material is considerably less devastating than formalin-fixed material on dermestids. (The fact that dermestids do not effectively clean formalin-treated specimens further discourages the use of dermestids for such material.) Preserving skulls in ethanol initially has the advantage of preventing damage caused by bacteria, fungi, insects, or other organisms. Skulls stored in a fluid medium are also free from problems associated with changes in relative humidity (Lafontaine and Wood, 1982; Plenderleith and Werner, 1971), and are somewhat buffered from temperature changes and mechanical damage. When specimens are prepared under adverse environmental conditions, ethanol preservation may be preferred.

Although there are some advantages to using ethanol there are also some potential problems such as the increased tendency for dermestids to damage tags of ethanol-treated specimens. *Dermestes* in confined conditions may forage on any available organic matter if dried flesh is not available. Dermestid damage to paper tags is often associated with specimen fluids impregnating the paper, thus the attractiveness of tags of fluid preserved specimens may be related to absorption of fats and oils from the ethanol. It may be necessary to seal the tag in a vial while the skull is being cleaned.

Based on the information presented above there seems to be justification for using ethanol for storing skeletal material in the field, particularly if drying conditions are unfavorable. However, there is a need to further investigate the possible effects such treatments might have on long-term preservation of the osseous tissues.

Conclusions

The effective use of dermestids for cleaning skeletal material relies on proper initial preparation. Standard drying of fresh or frozen specimens is appropriate for dermestid cleaning, but there is little benefit in removing the brains of smaller specimens. Skeletal material fixed in formalin should not be cleaned with dermestids. There are several benefits recognized with preparations involving only ethanol. Whatever method is used it is important that it does not compromise the integrity of the specimen being preserved.

Acknowledgments

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GOLDEN OLDIES: CURATING SEM SPECIMENS

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Abstract.—Literature on scanning electron microscopy (SEM) specimen preparation is extensive but permanent storage requirements for coated and mounted specimens are rarely mentioned. Uncoated specimens may produce acceptable SEM images, and it may be unnecessary to coat holotype specimens for SEM investigation.

SEM examination of specimens coated ten years previously shows no indication of the coating peeling off, and after the coatings were removed, the specimens showed no damage. Sturdy specimens may remain coated for long-term storage. Fragile microscopic specimens may require storage in vacuum desiccators. Institutions should include procedures for the use of specimens in SEM research in their curatorial and loan policies, and some suggestions for curatorial guidelines are presented.

Scanning electron microscopy (SEM) has been used for more than twenty years by biologists and paleontologists to reveal morphologic features. This technique yields images of such excellent quality that most systematic papers include SEM photographs. Thousands of coated specimens are being stored in repositories. What are the proper methods of storage and care for these specimens? The literature on SEM techniques and specimen preparation is extensive (Walker, 1978; Echlin, 1978; Murphy, 1982; Roomans, 1984; Robinson *et al.,* 1987), but permanent care and storage is not mentioned.

A review of current literature indicates that zoologists, botanists and anthropologists who describe ultrastructure and morphologic features consider their material expendable. The organisms or materials are plentiful and catalogue numbers are rarely cited. In contrast, paleontologists, and taxonomists in general, are very concerned about the preservation of type specimens. However, even the biological and paleontological literature contain only a few references that mention storage and preservation of SEM specimens, and most of these refer to microfossils, especially palynomorphs.

The lack of definite information on the storage requirements of research specimens is disconcerting. The original specimens have been altered by the coatings. Color patterns and internal structures are hidden, and in some cases, the coating may interfere with further examination by light microscopy. For non-type material, this may not be serious, but for primary type specimens, it is. In the future, a researcher may need to examine the original specimen. No one has investigated the long term effects of coatings on specimens or whether practical alternatives to permanent coatings exist.

I discussed these problems with SEM technicians, curators, and a chemist, and I tested some of the SEM and cleaning techniques on museum specimens. From this cursory inquiry, I have developed some general curatorial recommendations for specimens used in SEM investigation.

Uncoated Specimens

There are four requirements for any object to be investigated by SEM. The object must 1) be free of foreign particles and dust, 2) be vacuum stable, 3) remain stable after exposure to the electron beam, and 4) emit a sufficient number of

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secondary electrons and develop as few surface charges as possible (Robinson *et al.*, 1987:145). Most fossils and biological specimens fulfill the first three requirements without special treatment. It is the fourth item that causes problems; the specimens are not conductive. However, it is possible to photograph uncoated specimens in the scanning electron microscope. Moisture from a fresh specimen may damage the vacuum chamber but fossil and dry biological specimens do not pose this danger.

Fossil and Recent specimens of various taxonomic groups were selected from the general collections of the University of Iowa Paleontology Repository for this study. Both modern and fossil specimens are processed for SEM research in the same way and have similar curatorial problems. Soft tissue and medical specimens require special preparation techniques and are not considered.

To photograph uncoated specimens, the SEM must be set for low accelerating voltages and magnifications below 5,000×. Until about five years ago, scanning electron microscopes did not have the built-in option to produce low KV (the measurement of accelerating voltage). Uncoated specimens are run at 1–3 KV compared to 15–20 KV for coated specimens. Uncoated specimens have high resistance and uneven surface potential that cause the image artefacts called "charging" which can appear as bright spots or streaks on the photograph. Each specimen reacts differently to the exposure to the electron beam, and the degree of charging depends on a combination of specimen composition, size, surface relief, shape and magnification desired. An experienced, patient SEM operator must adjust and test the settings to achieve good results.

Paired photographs of three specimens (Figs. 1A–F), coated and uncoated, demonstrate that satisfactory images can be produced without coating specimens. The bright white areas on uncoated specimens (Figs. 1A, C, E) indicate charging. If, for the purposes of a study, a specimen can be photographed at lower magnifications (best under 2,000×, acceptable up to 5,000×), good quality SEM photographs that reveal morphologic features can be obtained without coating. At higher magnifications (Figs. 2A, C), strong charging is apparent but the images are clear. The coated diatom specimen at higher magnification (Figs. 2B, D) shows good surface detail but the uncoated specimen shows depth in the specimen (Fig. 2C). Clearly the coated specimens (Figs. 1B, D, F) yield better images, but if adequate SEM photographs of a holotype can be obtained for publication, then the uncoated option should be considered.

Institutions should seriously consider prohibiting researchers from coating primary type specimens for subsequent SEM investigation. Taxonomists should attempt photographing the holotypes of new taxa without coating them. Paratype and other secondary type material may be coated.

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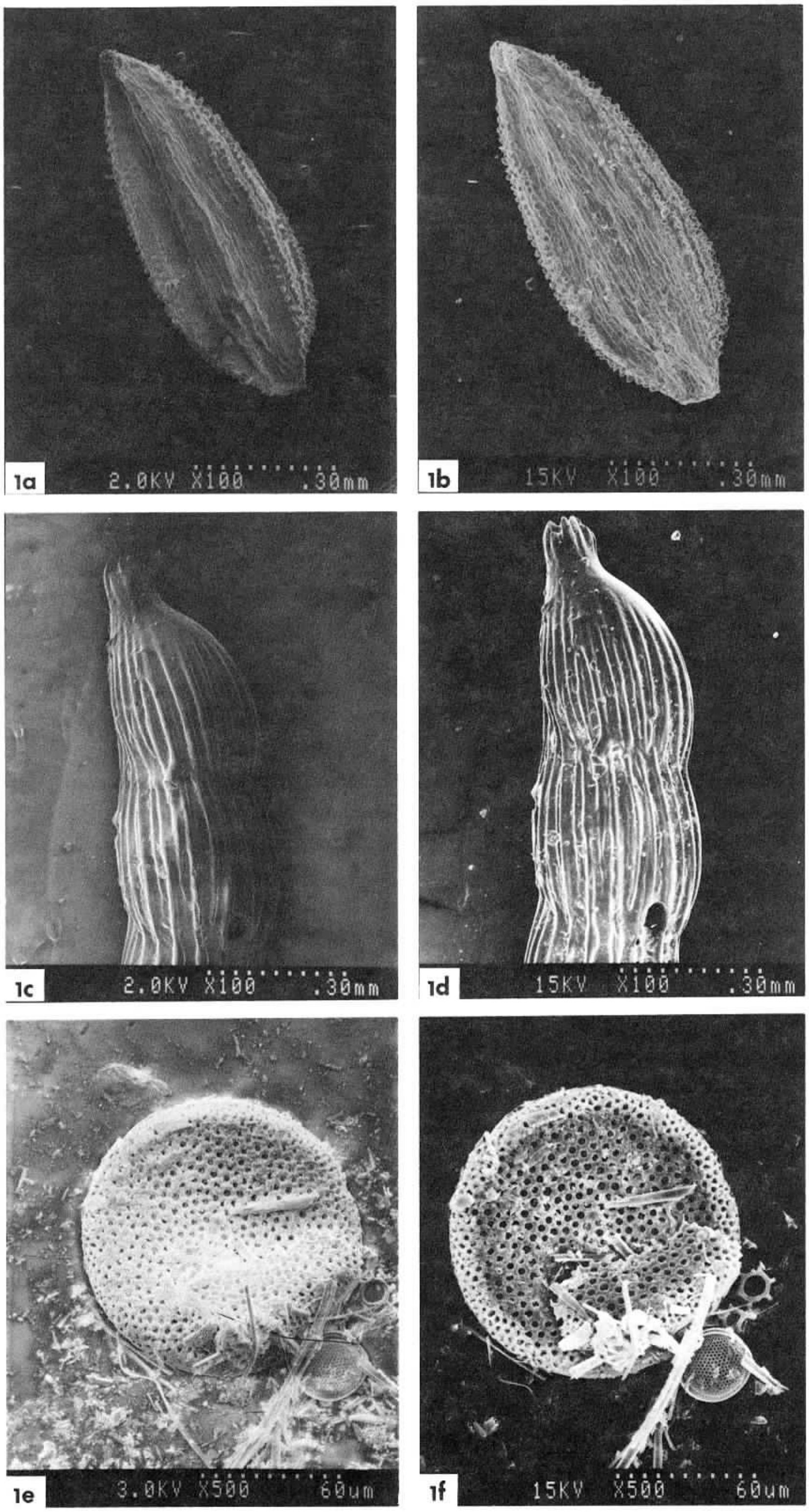
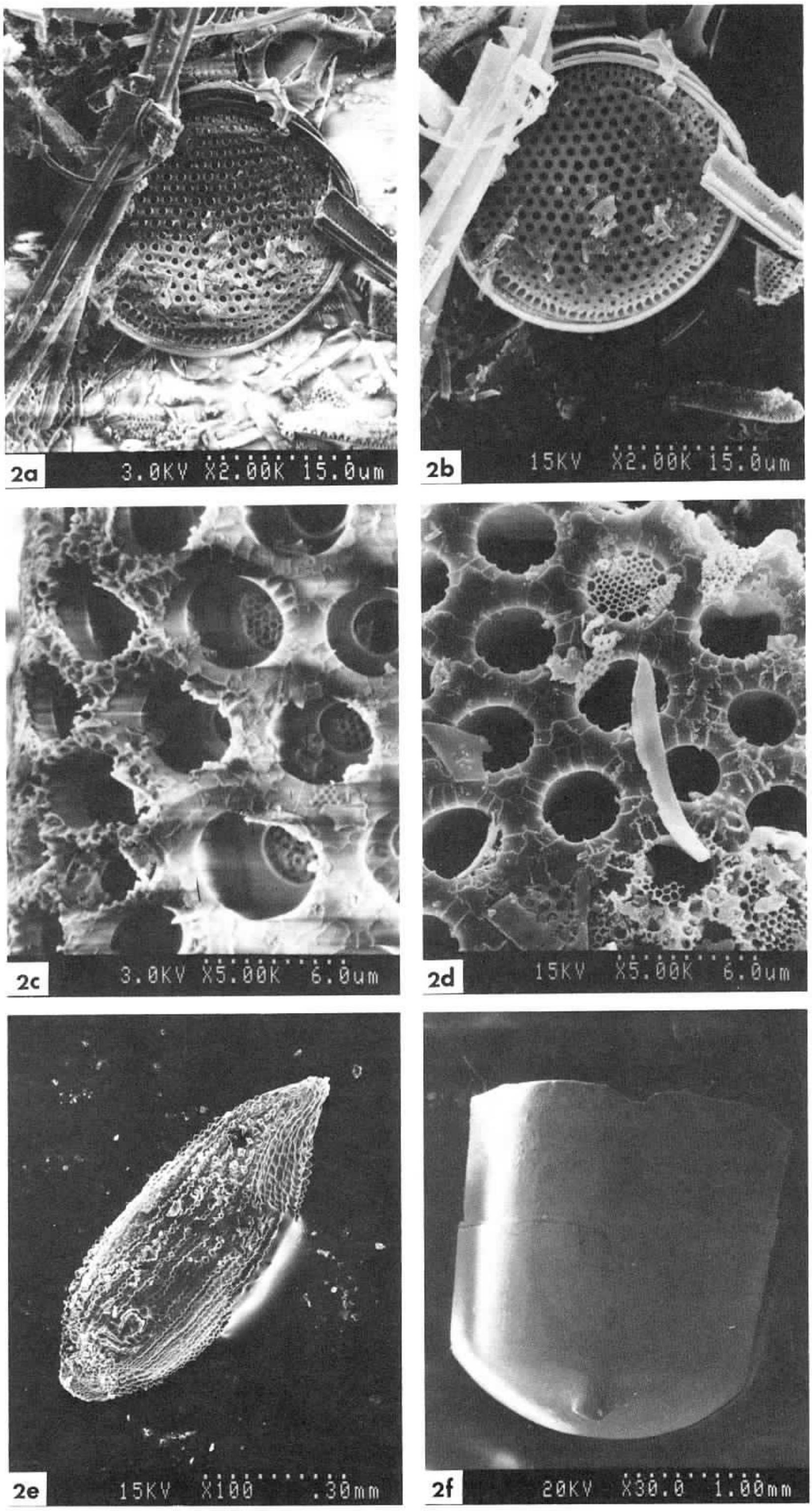
Figure legend. All photomicrographs in Figures 1 and 2 were taken with a Hitachi S-570 SEM on Polaroid positive and negative film PN55. The KV, magnification and size of the specimen are shown beneath each picture. The KV settings of 2 and 3 indicate uncoated specimens; the KV settings of 15 indicate coated specimens. The specimens are stored in the University of Iowa (SUI) Paleontology Repository.

Figure 1. a, b. Modern seed. *Epilobium glandulosum* (willow herb). SUI 54873A. *a.* uncoated. *b.* Au-Pd coated. c, d. Eocene unidentified nodosarid foraminifer, Alabama. SUI 54874. *c.* uncoated. *d.* Au-Pd coated. e, f. Modern unidentified marine diatoms. SUI 54875. *e.* uncoated. *f.* Au-Pd coated.


1a
2.0KV X100 .30mm
1b
15KV X100 .30mm
1c
2.0KV X100 .30mm
1d
15KV X100 .30mm
1e
3.0KV X500 60um
1f
15KV X500 60um


2a
3.0KV X2.00K 15.0um
2b
15KV X2.00K 15.0um
2c
3.0KV X5.00K 6.0um
2d
15KV X5.00K 6.0um
2e
15KV X100 .30mm
2f
20KV X30.0 1.00mm

Another alternative to coating specimens is to use a replica or cast. Replicas are necessary when the specimen is too large for the SEM chamber. The technique has been reported for fossil bone with great success (Bromage, 1987). The quality of current molding and casting materials provide excellent replicas or casts of the original surface without endangering the original specimen. For macrofossils and large specimens, replica or cast may be a viable solution for SEM photography of type specimens.

Coated Specimens

Coating a specimen has advantages. The variables that cause charging have no effect on a coated specimen and SEM photography is faster. Thin coatings are applied in one of two ways, vacuum evaporation or sputter coating. Many metallic and non-metallic materials may be used to coat a specimen. Experience has proven that of the metals, gold and gold alloys are the most practical because they are non-oxidizing, have small particle sizes, and have high sputtering yields (may be applied thinly). Most SEM facilities use gold or gold-palladium (Au-Pd) alloy.

A non-chemical technique to remove metallic coatings is to reverse the polarity of the sputter-coater. The coating is bombarded with Argon ions, and the loosened coating ions are attracted to the anode. This procedure is not effective. Coating the test specimens with Au-Pd took about 2.5 minutes; however, after five minutes of exposure to reversed polarity, no changes were visible on the specimens. An SEM operator at the Smithsonian Institution admitted that after thirty minutes of reversed polarity, the specimens he tried were not clean (Kahn, personal communication). Reversing the polarity is not an efficient use of the sputter-coater.

The most common method of removing the gold coating from specimens is immersion of the specimens in a cyanide solution (Hansen, 1968; Sela and Boyde, 1977). A major drawback is the toxicity of the cyanide solution. Precautions must be taken to perform the procedure in a fume hood, store the solution (labeled "POISON") in a fume hood, and dispose of the used solution properly. Sela and Boyde (1977) tested the cyanide procedure on fourteen different materials including organic tissues, skeletal and inorganic materials. Using scanning electron microscopy, the authors found no evidence of damage to any of the specimens. Urban (1968) reports a method for removing the gold coating from palynomorphs by immersing the specimens in aqua-regia before permanently mounting the specimens. This method is unsafe for most inorganic specimens.

Although the cyanide solution procedure is recommended for gold coating only (Sela and Boyde, 1977), in my tests, the cyanide solution completely cleaned the specimens coated with the gold-palladium alloy. Neither the SEM operators nor the chemist, to whom I spoke, were able to explain this (Dogan, Nessler and Eyman, personal communication). The specimens were clean and appeared unharmed.

Crissman and MacCann (1979) reported a procedure for gold-palladium removal using a solution of 10% $FeCl_3$ in ethanol. The procedure was sketchily

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Figure 2. a–d. Modern unidentified marine diatom. SUI 54875. *a, c.* uncoated. *b, d.* Au-Pd coated. e. Modern seed. *Epilobium glandulosum* (willow herb). SUI 54873B. Au-Pd coated. f. Pennsylvanian cephalopod, *Bactrites sinuosus* Mapes, Oklahoma. SUI 42564 Paratype. Gold-coated pre-1976.

described and their tests were run on Au-Pd coated Teflon tape only. This method needs testing because it may prove to be an inexpensive, safe method for removing the most common coating material Au-Pd.

Aluminum has been used as a coating metal for foraminifers (Sylvester-Bradley, 1969); however, it oxidizes very quickly. If the specimen will be photographed as soon as it is coated, and if the coating is removed immediately, then aluminum may be an option. Aluminum is removed by immersing the specimen in a dilute solution of sodium hydroxide for a few minutes.

A non-metallic material commonly used for coating is pure carbon. The Smithsonian Institution SEM Lab has used carbon on microfossils for years. The carbon is removed without chemical treatment by placing a specimen in a plasma asher. The carbon oxidizes rapidly and the specimens are unharmed (Kahn, personal communication). A similar method can be used for metallic coatings. Recently, the University of Iowa Central Electron Microscopy Facility purchased an Oxygen Plasma Etcher. Although the machine is not designed specifically to remove coatings, it may be used for this process. Under a vacuum, the metal coating is vaporized. I am currently evaluating this method, and if it proves to be efficient, reliable and cost effective, the etcher may become a safe, non-chemical treatment for removing metallic coatings from specimens.

Curatorial Considerations

Before specimens are coated for SEM, they must be completely dry and clean. Drying methods will depend on specimen composition and preparation techniques. After the specimens are coated, they should be stored in SEM stub holders in dust-proof conditions, preferably in a desiccator, until SEM examination is complete. Dust, moisture and other contaminants may damage the SEM vacuum chamber. Changes in humidity and exposure to air may affect the mounting medium and accelerate the deterioration of the coating. Following examination, permanent storage requirements are determined by both specimen composition and preparation history. For these reasons, before SEM work begins, researchers should confer with both the SEM technician, about selecting the proper preparation and mounting techniques for a particular application, and the curator, about permanent storage facilities.

Frequently, specimens are received for deposition mounted on SEM stubs without a record of mounting medium or coating material. One cannot always tell by sight what coating was used, but if a specimen is a bright gold color, then the coating is probably pure gold. Gold alloys and carbon appear more silver-colored. If the coating material is unknown, the composition can be determined by using the x-ray analysis option of the scanning electron microscope.

The adhesive used for securing the specimen to the SEM stub may affect the curation of the specimen as well as the quality of the SEM image (Fig. 2E, bright area shows the mounting adhesive on the specimen). Safely removing specimens from SEM stubs may pose a problem. The mounting medium must be dissolved without damaging the specimen. Witcomb (1981) examined 90 different adhesives including sprays, tapes, glues and epoxy adhesives, and evaluated materials for damage in the beam, drying times, ease of use, degree of tackiness, firmness and composition. Delicate microscopic specimens, attached directly to the SEM stub or dried on a flexible filter before attachment to the stub, are almost impossible to remove without harming the specimens. They must be stored on SEM stubs permanently, and perhaps in vacuum desiccators. More sturdy, larger specimens can be removed from the stubs and stored in the same manner as other specimens.

How to determine which specimens will require long term storage in vacuum desiccators has not been addressed in the literature and clearly needs further investigation. Two studies evaluated the storage and condition of specimens coated with Au-Pd for SEM. Small and Maugel (1978) examined protozoans they had freeze-dried, prepared and coated nine nine years previously. The specimens were stored in glass vacuum desiccators. Although the vacuums were not properly maintained, they were able to locate specimens previously photographed, and the specimens were in excellent condition. They attributed this to excellent preparation techniques.

In the other study, diatom specialists reexamined specimens, from culture, that had been prepared, coated with Au-Pd, and stored for four years (Lee *et al.,* 1986). Some stubs were stored in a vacuum desiccator and others were stored in a laboratory desk drawer in New York City. In SEM examination, specimens from both storage methods showed charging, and deteriorated coatings obscured the specimens. The authors feared that storage on SEM stubs either in dust-proof room conditions, or in a vacuum desiccator may not be an option. They strongly recommend that diatom type specimens be mounted for SEM on glass cover slips. After examination, the cover slips can be inverted onto standard microscope slides using a high contrast mounting medium for permanent storage. The thin gold layer will not interfere with subsequent light microscopy.

This method of inverting a glass cover slip onto a microscope slide for permanent mounting is used successfully by palynologists (Playford and Martin, 1984). However, the choice of permanent mounting medium must be made carefully. MacAdam (1971) reports that Clearcol mounting medium caused gold-palladium coatings to peel off the specimens and recommends Diaphane or Canada balsam for mounting.

I examined, by SEM, eight specimens (including fossil and modern corals, and fossils ammonoids and microcrinoids) that had been stored with both gold and gold-palladium coatings for more than ten years. Some of the specimens had been stored on covered microscope slides and the others were stored on SEM stubs uncovered in specimen boxes. All of the specimens had been stored in wooden cabinets that were not dust-proof. None of specimens showed any indication of the coatings peeling or flaking off. The gold coated specimens showed charging that indicated the coating had deteriorated (Fig. 2F); however, the specimens may be recoated safely for further SEM examination and will show no signs of charging. I removed the coatings from some of these specimens with cyanide and examined them. They showed no evidence of damage. For the very small sample I examined, it appears that neither the coating itself nor the removal of the coating harms the specimens. For most collections, uncoating every specimen is impractical. This preliminary study indicates that specimens may remain coated for long term storage.

Specimens can be damaged during shipment to the home institution, if proper precautions are not followed. Care should be taken to secure the SEM stubs in holders. Besides the stubs falling out of the holders, jarring may cause the specimens or cover slips holding specimens to pop off the stubs (the mounting adhesive must also be secure). A researcher may choose to curate the specimens personally by placing them permanently in storage boxes or on slides before shipping. More often however, the specimens are shipped on the stubs to be curated by the museum staff. Packing procedures for the return of material on loan should be spelled out in the loan agreement. In the case of new incoming material, suggestions for shipping can be made at the time a researcher arranges for deposition or when catalogue numbers are requested.

Recommendations

Because of the large number of variables in specimen composition, preparation techniques, mounting media, and coating materials, and the effect each has on storage requirements, specific recommendations will have to be developed by each specialized discipline. Institutions or each division within an institution should incorporate policies for specimens used in SEM investigations into their curatorial and loan statements. Some general guidelines are summarized in the Appendix.

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Appendix: Curatorial Recommendations For SEM Specimens

Coating Policy

1. Primary types (e.g., Holotype, Lectotype, Neotype)
   May not be coated (exceptions for unusual cases only; good quality photographs can be produced from uncoated specimens at magnifications under 5,000×)
2. Paratypes and secondary types
   May be coated with prior permission

Storage For Coated Specimens

1. Fragile specimens (especially microscopic specimens)
   Store on SEM stubs in plastic boxes designed to hold stubs and consider vacuum desiccator storage
   Or, permanently remount specimens on glass slides
2. Larger, more sturdy specimens
   Remove specimens from stubs (use correct solvent for mounting medium)
   Store coated in the collection in normal manner
3. All specimens
   Leave coating on (it is unnecessary to routinely remove coating*)
   Record coating composition, date coated and retain photograph
   Specimen may be recoated for subsequent examination if coating deteriorates

Preparation Policy

1. Request permission from curator to coat any specimen for SEM.
2. Researcher and SEM technician should discuss options for specimen preparation and mounting medium.
3. Record all specimen treatments including mounting medium and coating material (this information will be incorporated into the permanent record of the specimen).
4. Microscopic specimens may require special handling:
   a) mounting delicate specimens directly onto SEM stubs or flexible filters should be avoided
   b) mount specimens to be permanently stored on glass slides, individually on glass cover slips before coating
5. If coating will be removed, determine procedure and which institution will remove the coating.

Shipping

1. Specimens and cover slips holding specimens should be securely attached to SEM stubs.
2. Stubs should be secured in holders with neither the lid nor additional packing material touching the specimens.
3. Specimens should be clearly labeled (diagram multiple specimens on stubs).
4. Shock absorbing material should surround the stub holders in the packing box.
5. Type specimens should be sent by Insured or Registered Mail.

* It has been brought to my attention (Waller, personal communication) that the gold, gold alloy or carbon coating on a pyritized specimen may act as a cathode in an electro-chemical reaction (galvanic corrosion) and may cause serious damage to the coated specimens. It may be prudent to remove the coatings from pyritized specimens.

NATURAL HISTORY COLLECTIONS MANAGEMENT AT THE ROYAL ONTARIO MUSEUM

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Abstract.—Management of natural history collections at the Royal Ontario Museum (ROM) is the direct responsibility of each of ten Science departments, within the limits of institutional policies and procedures. The Co-ordinator of Collections Management makes recommendations to management on the best use of physical and financial resources to assure the welfare of the collections and co-ordinates cataloguing activities on the PARIS system of the Canadian Heritage Information Network.

Collections management is a ubiquitous term that means different things to different people. In the ROM's *Statement of Principles and Policies on Ethics and Conduct* (Royal Ontario Museum, 1982) collections management is defined as

> "the planned, organized, and co-ordinated effort of the institution to be accountable, at all times, for the collections it holds with respect to all physical and administrative aspects of their well-being."

Physical aspects include acquisition and disposal, treatment (including preparation, preservation, and conservation), and storage of collections and collections-related materials; administrative aspects include collections development policies and documentation. At ROM, flexible institutional policies allow different departments to respond to the traditions and needs of their various disciplines while being monitored by a minimum of controls imposed by the organizational structure of the museum.

The policies and procedures developed by an institution to manage collections and collections-related functions are very much a function of the history of the institution (Yamamoto, 1985). This is particularly well illustrated in the case of the ROM approach to collections management. Collections-related activities, although controlled by institutional policies, are developed in practice by the individual departments.

Figure 1 outlines the organizational structure of the ROM in 1988. The curatorial departments historically have been divided into two categories, Science, and Art and Archaeology, each with different approaches to collections management. When not on display, specimens and artifacts are located in the appropriate curatorial area, and, traditionally, responsibility for the collections has been assumed by the individual curatorial departments. Curators control the collections as they alone have the authority to initiate acquisitions and disposals.

Each of the ten Science departments controls its own documentation, with independent cataloguing and accessioning systems. Their systematic storage arrangements reflect the way the collections are used. Within the departments, collections management activities are usually the responsibility of a curatorial assistant, a professional with special experience and training (B.Sc. or M.Sc.) in the discipline. Arrangements for loans are made directly with the department

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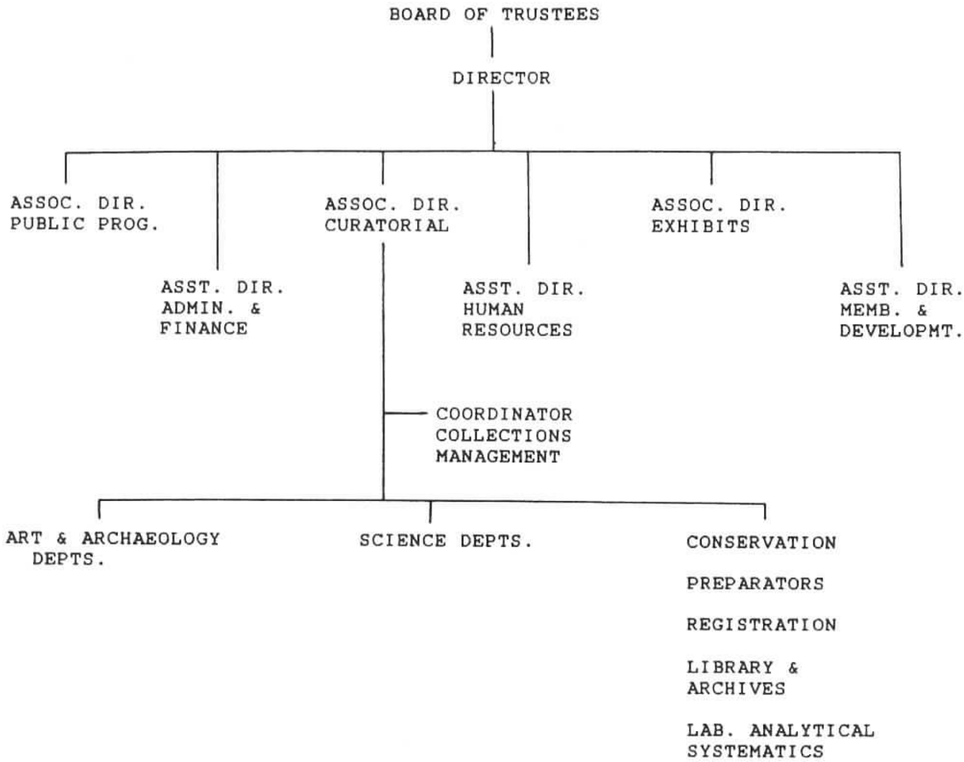

Figure 1. Organizational structure of the Royal Ontario Museum, 1988.

concerned. Criteria for loans and other uses of the collections are determined by the departments.

In contrast, the eight Art and Archaeology departments at the ROM are highly regulated, with a central catalogue registry for all departments and precisely defined procedures for all collections-related functions and activities. Treatment and documentation are carried out by or in conjunction with the curatorial service departments: Registration, Conservation, and Preparators. Indeed the activities of these service departments involve Art and Archaeology procedures almost exclusively, although they do act in an advisory capacity for the Science departments.

The organizational and procedural dichotomy has its beginnings in the early history of the museum. When founded in 1912, the ROM consisted of five museums, each with its own director: the Royal Ontario Museums of Geology, Mineralogy, Palaeontology, Zoology, and Archaeology. This imbalance in divisional structure formed the basis for the independent nature of the present Science departments. Through the years, the administrative structure changed. The five museums became four divisions: Earth Sciences, Palaeontology, Life Sciences, and Archaeology. The present structure of independent departments has evolved over the last 25 years or so. Thus one might say that the Art and Archaeology departments are monophyletic and the Science departments are polyphyletic. The evolutionary relationships are very obvious when collections management procedures are compared.

Overseeing this seemingly autocratic system is the Co-ordinator of Collections Management. This position was initiated in 1978 as ROM was beginning a major renovation and expansion project. As the project involved both new construction and the complete renovation of the old building, all collections had to be moved at least once. It was decided that whenever possible collections would not be moved off-site for storage during the construction and that the collections would remain accessible except during the actual move. The Co-ordinator of Collections Management was to oversee and co-ordinate all activities affecting the collections during this very disruptive period. A key criterion was that the person should be an existing staff member with a broad knowledge of the history of the museum and its collections management structure. Most importantly, it had to be someone who had the respect of the curatorial staff, the traditional "keepers of the collections," and could work well with them. The activities of the Co-ordinator of Collections Management during the renovation and move have been fully detailed by the incumbent (Yamamoto, 1985).

When the building construction was completed and the collections were installed in their new quarters, the position was re-evaluated and the value of a central facilitator was recognized. It should be noted that this position does not have authority over any collections, but rather makes recommendations to management on the best use of the museum's physical and financial resources to ensure the welfare of all collections. It is a staff position, reporting to the Associate Director Curatorial.

The duties of the Co-ordinator of Collections Management include many functions previously covered by other departments or individuals. The Co-ordinator manages the pest control budget (previously handled by the Conservation Department) and co-ordinates all pest control programs within the museum including routine and special fumigations of areas and objects. He co-ordinates risk management projects as they pertain to the collections (Yamamoto, 1988). This includes continuous upgrading of collection storage areas, ensuring adequate space and equipment to store collections safely, and identification of situations that might put collections at risk (e.g., proximity of sprinkler heads and other building services, vibration, environmental control, etc.). In this capacity he works closely with the curatorial departments and with the Security and Physical Plant Departments. He has been instrumental in the development of a comprehensive disaster plan for the museum.

Administratively, the Co-ordinator of Collections Management chairs the Collections Management Committee, which helps co-ordinate the physical and human resources for collections management. The committee is made up of representatives of the primary users of collections within the museum and the personnel responsible for handling, treatment, and documentation of the collections. Although most activities with which the committee is concerned relate to art and archaeology collections, certain issues do impact on science collections as well.

The office of the Co-ordinator of Collections Management, consisting of an assistant, technician, and data entry clerk, also co-ordinates documentation through the computer cataloguing activities of departments on the PARIS system of the Canadian Heritage Information Network (CHIN) (Cox, 1986). The Registration Department, which co-ordinates all art and archaeology collections administration, went on-line with CHIN in 1978 with a location record project to keep track of art and archaeology collections during the expansion project. The departments of Textiles and Ornithology were already on the system, having been involved since 1976 in a pilot project with the National Inventory Project (NIP), precursor to CHIN.

Several Science departments were using an in-house computer cataloguing system started in 1971. Starting in 1982, the departments of Ichthyology and Herpetology and Invertebrate Palaeontology started entering records on the Natural Sciences Data Base of CHIN, and their in-house data files were also transferred to the PARIS system. At the same time the Vertebrate Palaeontology Department started inputting records. There are now seven Science departments on-line with CHIN with a total of over 287,000 records. Each department has its own file within the Natural Sciences Data Base. The structure of the different files takes into account the different management methods of the disciplines.

Although ROM has ten separate departmental files within two CHIN data bases, it is considered a single user by CHIN. CHIN provides the line feed and a multiplexer to communicate data to its mainframe. The Co-ordinator of Collections Management supplies participating departments with the necessary hardware and dataline resources to access the multiplexer. The office of the Co-ordinator of Collections Management also provides advice on hardware and software questions and occasional personnel assistance in inputting and editing on a project basis.

Although departments can work independently on-line with CHIN for data entry, retrieval, and manipulation, several departments prefer to do bulk entry and editing of files off-line using a microcomputer. Batch files are then loaded to or from CHIN through the office of the Co-ordinator. Thus although the departments are independent from one another in their documentation, they are to varying degrees dependent on the Co-ordinator of Collections Management for some of their cataloguing activities.

The ROM system of collections management provides a workable balance of departmental independence and institutional accountability. Maintenance of the collections is performed by discipline specialists who know their collections and are aware of the special needs of both the collections and their users. As most science acquisitions involve field work, the staff involved are responsible for obtaining their own permits for collecting, export, and import of materials. Before the advent of the Co-ordinator of Collections Management individual departments were responsible for seeing to all the needs of their collections alone. The Co-ordinator of Collections Management acts as an impartial advisor to help ensure that all collections receive the best possible care available within the financial and human resources of the museum.

Proposed Changes

In February 1989, the Board of Trustees approved implementation of a new Strategic Plan that incorporates several structural administrative changes. This plan calls for the formation of a new Department of Collections Management to include the present departments of Conservation and Registration and the office of the Co-ordinator of Collections Management as well as the Photography section, presently administered by Exhibit Design Services. The inclusion of photography will facilitate the photodocumentation aspect of collections administration. With the redefinition of the major objectives of the museum to include installation of new galleries as a high priority, the preparators will now be administered from the Exhibit Design Services Department, reporting to the Associate Director Exhibitions. The Library and Archives will temporarily report directly to the Director.

Acknowledgments

I am grateful to Tosh Yamamoto for many long discussions and his critical input into this paper.

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A TREATMENT FOR BISON HORNSHEATHS

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Abstract.—Archaeologists and paleobiologists have a long tradition of borrowing and trading field techniques for conservation of artifacts and specimens. Over a period of several field seasons in Labrador, a treatment for wet archaeological baleen was developed. This caught the attention of a conservator with the National Museum of Natural Sciences (NMNS), Ottawa, who saw this as a possible treatment for a large collection of bison hornsheaths, which are similar structurally to baleen and had been collected under similar field conditions. This paper discusses the treatment developed for the bison hornsheaths.

A number of hornsheaths in the NMNS collection, dating to between twenty or thirty thousand years B.P., were washed out of frozen mud by placer miners in the Dawson area of the Yukon Territory. They were left on the surface until collected during the summer field seasons by paleobiologists. Their condition when excavated is not known, but by the time they were collected, many were badly desiccated and had the appearance of "exploded cigars" (Fig. 1). Presumably these hornsheaths had been through a number of freeze/thaw, wet/dry cycles.

The importance of many of these pieces for paleobiologists lies in their use as samples for carbon 14 dating and other possible forms of archaeometry. That they are in a badly desiccated state will not affect the results, in fact, field treatments are discouraged in case they jeopardize future analytical work.

There are other hornsheaths in the collection, however, which could enhance the knowledge of these ancient animals if they were conserved. In particular, these are hornsheaths as part of entire skulls and fitted on the boney horn core. If these structures could be conserved it would enable closer study of their gross morphology. Scientific measurements can be taken for projections for artistic reproduction, reconstruction or to add to scientific data. The original specimens would also be hardy enough for display.

Keratinous materials including hair, beak, hoof, feather, horn, and baleen to name a few, are all basically composed of nitrogenous substances, and all react to heat and moisture in similar ways. One of the treatments performed on baleen between 1978 and 1982 was the application of moisture and consolidant on badly delaminated specimens. When they were immersed in warm water it was found that almost immediately the delaminating and warped pieces relaxed, and became quite supple. Similar satisfactory results were achieved through immersion of the object in Rhoplex AC-33®, an acrylic emulsion, and when pieces were held in place with weights through a slow drying period, the realigned layers of keratin stayed in place. After several years of storage at 40% relative humidity the realigned layers of horny tissue have remained unchanged.

After some discussions and examinations of the bison hornsheaths in the Paleobiology Division of the National Museum of Natural Sciences, it was decided to test some bison horns to determine if they would respond to this treatment in a manner similar to baleen. Several small samples averaging about 5 cm × 1.5 cm × 0.2 cm were removed from specimens (NMC numbers: 21072, 40261, 402633, 11782, 37681, 11669, 35162, 11785, 36389, 25170, 363,42, 33877) and

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Figure 1. NMC 21072: Specimen showing typical delamination pattern.

were tested using the same technique as that used on the baleen. They reacted favourably to the treatment. But how would larger, and thicker samples respond? The more robust specimens would also have more strength and "memory." The problem of pieces reverting to their "exploded cigar" appearance could occur if they were not sufficiently restrained.

Initial Treatment

Four hornsheaths were treated in succession (NMC numbers: 37632, 37621, 13518, 21072), to determine the mechanics of the treatment. The procedure included the following: a) immersion in warm water for lengths of time from one to sixty minutes, b) reshaping specimen, c) consolidation with 2–5% solutions Rhoplex AC-33 and binding simultaneously, and d) slow-drying over a period of one to four weeks.

Some common working qualities of the horn became apparent. Although these pieces were extremely fragile, and delaminated at the slightest touch, they still retained a "memory" and required strong binding during the drying process to prevent them from warping back to the shape they had acquired during or after burial. The insides had to be wedged with pieces of inert polyethylene foam (Ethafoam®) to keep the hornsheath from collapsing on itself under the stress of the binding. The bison-horn reacted so quickly to the drying process that one could not leave it to air-dry without the constraints for more than fifteen or twenty minutes before the sheets of horny tissue would start to bend up.

Parafilm, a stretchy laboratory film was used to bind some of the specimens. Parafilm is manufactured by American National Can Corporation, and is a poly-isobutylene film with polyethylene/paraffin hydrocarbon wax surface coatings. It

Figure 2. NMC 11782: Untreated specimen on bone core.

provided the necessary strength and flexibility but because it is impermeable to moisture, the specimens dried only when the Parafilm was removed. The slippery adhesive coating made handling the pieces awkward and once dry, the consolidant was difficult to remove. The drying process was labour-intensive, as sections of the Parafilm had to be removed daily for controlled air-drying. The final results were quite satisfactory, in that the pieces were consolidated, but two of the hornsheaths exhibited a surface gloss due to excess surface consolidant.

Specialised Treatment

As the initial treatments were so successful, it was decided to attempt a more complicated specimen. This was a large (53 cm long) hornsheath (NMC 11782) which was still on the bone core (Fig. 2).

Preparation of this complete specimen for display involved new concerns. Foremost, the horn-core and hornsheath could not be immersed as a unit in a hot water bath. It would be impossible to realign the hornsheath while still on the core, and since the core was stable it would be foolhardy to soak it in water. If they were separated it was improbable that the hornsheath could be reshaped to fit back properly on the core. The solution was to remove the hornsheath and make a polyesther/fibreglass cast of the bone core to permit immersion of the sheath without damage to the bone (Fig. 3)

In order to ensure that the hornsheath could be placed back on the horncore after treatment, the cast was inserted into the hornsheath. Both the hornsheath and cast were then immersed in hot water. Due to the thickness and robust nature of this hornsheath compared to the others previously treated, hot water was used in the bath. The hornsheath and cast remained in this bath for about an hour,

Figure 3. NMC 11782: *Left*, Bone core; *centre*, fibreglass/polyester mold; *right*, hornsheath.

being periodically checked for flexibility. When it was sufficiently flexible to be reformed, it was removed. It would have, perhaps, been easier to work with had it remained longer in the waterbath but there was a concern that if the hornsheath became too wet it might become glutinous. A dilute solution (1–5%) of Rhoplex AC-33 was then brushed on the surface. The consolidant was injected under lifting layers and quickly bound with Parafilm® or clamped. The next day, as sections of parafilm were replaced, excess adhesive that had accumulated on the surface was removed with a stiff stipple-brush dipped in hot water.

The main criteria for a suitable binding agent for the drying process were as follows:

1) It must not adhere to the artifact or to itself when used with the consolidant.
2) It must be stretchy enough to accommodate irregularities in the horn surface, yet at the same time strong enough to apply uniform pressure around the circumference of the object.
3) It must be easy to use by the conservator working alone, and inexpensive so that it can be disposed of after each use.

There was some experimentation with Gortex® a random-pore teflon product and Reemay® a random-spun polyester. Although both products have good release properties they were not stretchy enough to apply the pressure required in a binding agent. A rather unorthodox binding agent was then tried: strips of pantyhose (100% Nylon) satisfied all the requirements and worked equally well with or without release paper. Drying took place over a period of four weeks. Another advantage to using these nylon strips was that air drying was accomplished without the need to periodically remove the binding.

Figure 4. NMC 25169: Specimen during drying process, wrapped with Reemay and bound with panty-hose.

Even after care was taken to avoid buildup of excess consolidant, after drying there were still some shiny spots on the finished hornsheath which were difficult to remove. Although the actual treatment had been worked out, the problem of excess adhesive still remained. It was observed that when the bindings were removed prematurely, the horn started to revert back to its pre-treatment shape. This demonstrates that very controlled drying conditions are necessary and it may not be possible or desirable to completely restrain some robust pieces.

A stronger adhesive would not provide an acceptable solution. If the adhesive is too strong the layers of the hornsheath will themselves separate before the adhesive fails. (The use of a bond that is weaker than the substrate is a standard sought in conservation.)

In an attempt to eliminate the accumulation of excess adhesive, the treatment was modified. The specimen which was selected (NMC 25169) appeared to be in a condition as degraded as the pieces which had been previously treated. It was immersed in hot-water until the delaminating layers realigned. Some pieces which were separate were placed in their appropriate position as the consolidant and bindings were applied. Dilute (about 5%) Rhoplex AC-33 was applied by brush and pipette to sections of the horn. The excess was removed with a dampened sponge, and the horn was wrapped with Reemay and bound with panty-hose. This was then rinsed under warm water and allowed to drain. The specimen was left bound and allowed to dry for several weeks (Fig. 4).

Conclusion

The visual results of these treatments are very favourable (Fig. 5). In conservation practise, the rehydration of a desiccated object is quite unusual. Usually,

Figure 5. NMC 11782: After treatment: hornsheath fitted onto bone core.

items from wet sites are kept wet until treated, and items from dry sites are kept dry. In the case of these hornsheaths an experimental treatment was required because there simply were no alternatives. If the specimens remained unconsolidated, they would continue to loose flakes and larger pieces whenever they were handled.

Rhoplex AC-33 is quite alkaline when wet and therefore its use on protinaceous material has been questioned (Down and Williams, 1988). For this reason care was taken to start out with small sample pieces, and when treatment was performed on larger specimens, a minimal amount of consolidant was used. Another Rohm & Haas acrylic worth consideration as an alternative to Rhoplex AC-33 is the colloidal dispersion, "Acrysol WS-24," which has been gaining recognition as a good consolidant for bone. It has a neutral pH and a higher glass transition temperature. Its fine particle size should theoretically allow for better penetration, but for repairing splits in delaminating layers there may not be sufficient tack.

Although specimens of baleen treated with Rhoplex® have remained stable ten years after treatment, the stability of the hornsheaths can only be observed after many years as well. Analysis has not yet been carried out to determine any damage at the microscopic level. The bison hornsheaths are, however, much less fragile in the consolidated form, thus making display possible and permitting handling for study and research.

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Down, J. L., and R. S. Williams. 1988. Report on adhesive testing at the Canadian Conservation Institute. *Handout,* The Conservation of Historic and Artistic Works on Paper, "Symposium '88," Canadian Conservation Institute, Ottawa.

BOOK REVIEWS

MAMMAL COLLECTION MANAGEMENT, 1987, H. H. Genoways, C. Jones, and O. L. Rossolimo, eds. (Texas Tech University Press, Lubbock, 219 pp.). *Mammal Collection Management* consists of 17 papers presented at a workshop sponsored by the International Commission for Mammal Collections during the Fourth International Theriological Congress, Edmonton, Canada, 1985. The volume is divided into three parts: specimen preservation and utilization, the employment of computers, and international problems and perspectives. Yates' introductory article documents the value of specimens and collections in systematic, biogeographic and speciation studies as well as the variety of emergent and traditional disciplines dependent upon such materials. The ensuing section considers aspects of specimen preservation and utilization. Williams and Hawks provide an outstanding compilation of literature on the history of preparation materials since the 1600s, exclusive of tanned skins and skeletal materials. Jones and Owens recommend specific procedures in curation, fixation, washing, storage and maintenance of fluid preserved specimens. In a more applied, yet enlightening review, George discusses the detection of environmental contaminants in mammalian material.

The large middle section of the volume considers the selection, uses and employment of computers. Williams reviews the criteria for the selection of computers, aided in part, by the responses of curatorial staffs at major museums. McLaren, Genoways and Schlitter demonstrate time-saving, collection management techniques in their employment of computers: taxonomic/geographic updates and inquiries, collector number searches, loans and accessions, cross reference files, specimen labels, etc. Following this is a description of data processing procedures in past or current use at four major North American Museums: Smithsonian (mainframe), Field Museum (minicomputer), Royal Ontario Museum (minicomputer) and Texas A&M (minicomputer/mainframe hybrid). This section demonstrates the unmatched benefits of computerization and is especially useful for those institutions in the process of specimen computerization or considering the variety of options available.

International collection concerns are addressed in the final segment. Of more general concern and appeal are the two articles dealing with problems in 'Non-Western' countries. Agrawal and Chakraborty discuss the maintenance problems (and ad hoc solutions) of a natural history collection in the tropical environs of Calcutta. These include climate, storage, space, light, pollution, pests, temperature and humidity control, etc. Pefaur surveys many Latin American collections and provides excellent data on the number and size of collections, qualifications of professional staff, housing, funding, etc. In this regard a review of the means by which Western European institutions are coping with severe budget cuts (e.g. BMNH) would be most interesting. In the United States, ever growing collections and use of natural history depositories has dramatically increased without increases in professional staff. The need to prioritize curatorial efforts in Western institutions is not addressed in the symposium. A few of the articles are not of general significance for the management of mammal collections. Despite this, the level of problem solving reflected in many of the contributions makes the acquisition of this volume a priority for any expanding and developing collection.—

Collection Forum, 5(1), 1989, pp. 38–40

Julian C. Kerbis, Division of Mammals, Field Museum of Natural History, Chicago, IL 60605-2496.

A CONSERVATION MANUAL FOR THE FIELD ARCHAEOLOGIST, 1987, Catherine Sease (Institute of Archaeology, University of California, Los Angeles, 169 pp.). The purpose of the manual is clearly stated in the introduction: "This book is aimed at the field archaeologist. Its purpose is to provide excavators with the basic conservation techniques necessary to safeguard and protect artefacts from the moment they are unearthed until they are fully treated in a properly equipped conservation laboratory by professional conservators."

Although it is intended for archaeologists, it should also be of use to professional conservators who have to deal with excavated material or who are going into the field for the first time. There has long been a lack of comprehensive English language texts on field conservation with the exception of David Leigh's "First Aid For Finds" (recently updated by David Watkinson and reissued in 1987), and Elisabeth Dowman's "Conservation in Field Archaeology" (1970), which has been out of print for many years.

Sease is well qualified to produce such a manual having had practical first-hand experience on archaeological excavations in many parts of the world. Her familiarity with field conditions and the need for flexible thinking and innovative responses to the unexpected is evident throughout the book.

The main tenor of the manual is directed towards European, Middle Eastern and Southern American archaeology, especially in dry regions, on sites far removed from the home base. It delves less deeply into the special conditions pertaining to tropical, arctic, or underwater sites, nor does it deal with many of the modern materials (e.g., tin, aluminium, etc.) likely to be encountered in industrial archaeology.

The first chapter covers several essential topics: the effects of excavation, the importance of leaving artefacts untreated whenever possible, the principle of reversible treatments, and the danger of cleaning and conservation treatments that can invalidate any subsequent analysis. It should be written in blood.

The second chapter deals with safety. It is very refreshing to see this topic given a prominent position in the text instead of being placed in an appendix as an afterthought. Some essential comments are made about transporting, storing and disposing of chemicals with the useful reminder that empty containers with chemical residues might be reused unsuitably by the local population unless they are made unusable before they are discarded. There is also a short section on first aid and cleaning up spills in case of accidents with the conservation chemicals. Both the spills and the disposal of chemicals rely on the availability of copious amounts of water. Sease warns against storing or mixing incompatible chemicals together and there is some information about inadvisable combinations scattered through the text, but for field personnel who are not chemists perhaps a chart of the incompatible chemicals likely to be in use on site, could be inserted into future editions.

Chapter three and Appendix III deal with supplies and suppliers. Some of the descriptions of the chemicals and consolidants are disappointingly superficial. For example, ammonia: no information is given about its pH, its flammability or its incompatibility with nitric acid (except by inference because it is classed in this book as a solvent). Nor is there enough discussion of the distinctive individual properties of the various solvents and resins she has selected, to make it clear to the non-conservator when it would be preferable to use one rather than another. Several fungicides are recommended for wet material with due warning about the hazards. However, in view of the difficulty of using and disposing of them on site, (Sease suggests small amounts can be thoroughly diluted and flushed down the drain or dumped in a deserted area), it is surprising to see so little reference to less toxic alternatives. She does briefly mention refrigeration with its attendant need for supervision to prevent freezing, but not ethanol or isopropanol which are quite commonly used in the field.

Packing, record keeping, block lifting, consolidation and other general treatments are discussed in the fourth chapter, the fifth being devoted to the identification and treatment of specific materials. It is clear that the reader is expected to be familiar with the substance of the earlier chapters before carrying out the instructions in chapter five. Although Sease has explained that the treatments are meant to be first aid methods and that they may be "the only treatment the objects will ever receive" (p. xi) in some cases they might be putting the artefact at risk for purely cosmetic reasons. For example, "If the ivory is in very good condition, surface dirt can be removed with swabs dipped in water. . . . If the ivory shows any signs of cracking or warping—which can happen before your eyes—stop . . ." (p. 80 and 81). The same might be said for consolidation which is suggested for many materials in spite of the excellent disclaimers in chapter four.

There is a tendency for information to be given in the form of statements often without discussion. As the instructions are directed towards non-conservators, it would be helpful if there was a little more explanation to support them. Even conservators might wonder why Sease insists that "Ammonia . . . is used only as a solvent for rubber latex" (p. 14, §5); "Panacide, also called Dichlorophen, can be used for wood and basketry, although only the disodium salt should be used" (p. 19, §7); "Amber is not brittle" (p. 50, §2); "It is not necessary to use silica gel when packing lead" (p. 83, §5).

There is an error in Appendix II which describes how to make up solutions. Having correctly said that the concentration of a solution is expressed as the amount of solid per unit volume of solution, Sease then gives step by step instructions which fail to account for the displacement of the liquid by the solid. In many cases it would not matter, but for acid stripping agents or biocides, accuracy is desirable in order to keep the concentration to an absolute minimum.

In spite of these drawbacks the manual has a great deal of useful information. The text is clear and well organised, avoiding technical jargon, and important points are highlighted in the margins. It is generously illustrated throughout with diagrams and useful tables, as well as 46 black and white plates at the back illustrating items such as the texture and characteristic appearance of specific materials. Although it is primarily aimed at archaeologists the sections on packing, handling, safety, supply sources, block lifting and taking impressions, etc. may also be of interest to palaeontologists.—*J. Fenn, Conservation Department, Royal Ontario Museum, 100 Queen's Park, Toronto, ON M5S 2C6.*

Preparation of Manuscripts

General.—It is strongly recommended that, before submitting a paper, the author ask qualified persons to appraise it. The author should submit three copies of the manuscript either typewritten or printed on letter quality printers. **All parts of the manuscript must be double spaced** with pica or elite type on 8½ × 11 inch (21.6 by 27.9 cm) or A4 paper and at least one inch (2.5 cm) margins on all sides. Manuscripts should not be right justified, and manuscripts produced on low-quality dot matrix printers are not acceptable.

Each page of the manuscript should be numbered. Do not hyphenate words at the right-hand margin. Each table and figure should be on a separate page. The ratio of tables plus figures to text pages should generally not exceed 1:2.

The first page includes the title of the article, names of authors, affiliations and addresses of authors, and the abstract if present. In the top left-hand corner of the first page, indicate the name and mailing address for the author to whom correspondence and proofs should be addressed. All subsequent pages should have the last names of the authors in the upper left-hand corner.

The preferred language for manuscripts is English, but a summary in another language can precede the literature cited, if appropriate. Manuscripts written in other languages will be considered if the language uses the Roman alphabet, an English summary is provided, and reviewers are available for the language in question.

Abstract.—An abstract summarizing in concrete terms the methods, findings and implications discussed in the paper must accompany a feature article. The abstract should be completely self-explanatory and should not exceed 200 words in length.

Style and abbreviations.—Symbols, units, and nomenclature should conform to international usage. Cite all references in the text by the author and date, in parentheses. Footnotes should be avoided. For general matters of style authors should consult the "Chicago Manual of Style," 13th ed., University of Chicago Press, 1982.

Literature cited.—This section includes only references cited in the manuscript and should be typed double spaced. References are listed alphabetically by authors' names and take these forms:

Jones, E. M., and R. D. Owen. 1987. Fluid preservation of specimens. Pp. 51–64 in *Mammal Collection Management* (H. H. Genoways, C. Jones, and O. L. Rossolimo, eds.). Texas Tech University Press, Lubbock, 219 pp.

Sarasan, L. 1987. What to look for in an automated collections management system. Museum Studies Journal, 3:82–93.

Thomson, G. 1986. *The Museum Environment,* 2nd ed. Butterworths, London, 293 pp.

Tables and illustrations.—Tables and illustrations should not repeat data contained in the text. Each table should be numbered with arabic numerals, include a short legend, and be referred to in the text. Column headings and descriptive matter in tables should be brief. Vertical rules should not be used. Tables should be placed one to a page, after the references.

All figures must be of professional quality as they will not be redrawn by the editorial staff. They may include line drawings, graphs or black and white photographs. All figures should be of sufficient size and clarity to permit reduction to an appropriate size; ordinarily they should be no more than twice the size of intended reductions and whenever possible should be no greater than a manuscript page size for ease of handling.

Photographs must be printed on glossy paper, with sharp focus and high contrast essential for good reproduction. Photos should be trimmed to show only essential features.

Each figure should be numbered with arabic numerals and be referred to in the text. Legends for figures should be typed on a separate sheet of paper at the end of the manuscript. Magnification scale, if used, should be indicated in the figure by a scale bar, not in the caption. Notations identifying the author and figure number must be made in pencil on the back of each illustration. All illustrations must be submitted as an original and two copies. Note placement of tables and illustrations in the margins of the manuscript.

Evaluation of a manuscript.—Authors should be aware that the following points are among those considered by the editorial staff when evaluating manuscripts: 1) Is the content appropriate to the purpose of the journal and society? 2) Are the contents clearly and logically presented and the paper well organized? 3) Is the methodology technically and logically sound? 4) Does the paper contribute to the body of knowledge and literature? 5) Is the study integrated with existing knowledge and literature? Is the literature cited appropriate for the study? 6) Are the conclusions supported by sufficient data? 7) Does the title reflect the thrust and limitations of the study? 8) Are the tables and figures clearly presented? Are they necessary to support the text?

Submission Procedure

Manuscripts intended either as feature articles or general notes should be submitted in triplicate (original and two copies) to the Managing Editor. Letters to the Editor and correspondence relating to manuscripts should be directed to the Managing Editor. Books for review should be sent to the Associate Editor for Book Reviews.

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