NOTES

from a

WORKSHOP ON BIRD SPECIMEN PREPARATION

held at

THE CARNEGIE MUSEUM OF NATURAL HISTORY

in conjunction with the 107th Stated Meeting

of the

AMERICAN ORNITHOLOGISTS' UNION

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Compiled by

STEPHEN P. ROGERS

and

D. SCOTT WOOD

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INTRODUCTION

The use of museum collections has expanded considerably in scope over the past several decades. Such collections, once used primarily in systematic or faunistic studies, are now also commonly consulted in conjunction with ecological, biochemical, behavioral, and other types of research. Several novel preparation techniques have been developed during recent years, often in response to these non-traditional uses. Unfortunately, the training of students in museum techniques has been greatly reduced. Not only are fewer museum technicians being trained, but very few students of ornithology (or of any area of biology) receive so much as an introduction to even traditional museum methods, let alone those of more recent development. Even those students interested in research careers in museums are often poorly educated in the techniques of collection management and specimen preparation.

A compounding problem is the lack of inter-museum communication between staff members responsible for the care and management of collections: Scientific Preparators, Curatorial Assistants, and Collection Managers, (and, to a lesser degree, Curators). The creation of the Society for the Preservation of Natural History Collections (SPNHC) has finally provided a professional forum for these workers, but many museum administrators still view sub-curatorial staff as something less than professional and, consequently, do not make the same kind of travel and other resources available to them as enjoyed by the curators.

We had two primary purposes for this Workshop. First, we wanted to assemble in one spot as many people interested in bird preparation as possible so that they could get to know one another--Communication over distance is much easier among acquaintances and friends. Second, we wanted to highlight several of the recent developments in bird specimen preparation so that this information would be available to the museum community. The compilation of the papers presented allows a permanent record which can be repeatedly referred to, and permits dissemination of this information to those workers throughout the world who were unable to attend the workshop.

We were very pleased with the results of the Workshop. Nearly 100 persons attended, including a considerable number who would not otherwise have attended a national meeting. The papers presented were interesting, provocative, and entertaining, and will be a base for future efforts. We hope this Workshop will be the first of a long series of such events held in conjunction with meetings of both the AOU and SPNHC, as well as other suitable organizations.

We thank The Carnegie Museum of Natural History for providing the space for the Workshop, and also the members of the American Ornithologists' Union Committee on Local Arrangements for their help, encouragement and support. We extend special thanks to the twelve presenters and to Ms. Christine Skelly for her audiovisual help.

Stephen P. Rogers
D. Scott Wood
Carnegie Museum of Natural History
1 October 1989

Workshop on Bird Specimen Preparation

Schmoo Preparation

Robert W. Dickerman

American Museum of Natural History, New York, NY

and

Museum of Southwestern Biology, Albuquerque, NM

Many ornithologists have been preparing flat skins with skeletons for years. In most cases, these have been for specific studies such as molt, plumage, and pterylosis, or in order to have a skeleton and yet have information useful for taxonomic and distributional studies. However, as valuable as they are for certain studies, flat skins are less than ideal for making color comparisons with round skins. Schmoos may be completely boneless with a complete skeleton preserved, may have one measurable leg and wing left on the skin, or may have the bill cut off just inside the skin so that greater than 90% of the skeleton is preserved. I will first describe the methodology of schmoo preparation in a large bird and will then mention adaptations useful for small birds.

To begin, an incision is made from approximately one inch above the end of the sternum to the anus, and skinning starts in the traditional manner. Each leg is exposed and cut at the

distal end of the femur. If tarsi are feathered or tarsal scutulation is important, one leg may be left on the skin. One leg, usually the left, is disarticulated at the distal end of th tibia. The tibia of this leg is later replaced with a wrapped stick. On the other 81eg, I slit the tarsus up the back, extending the cut about one-fourth of the way up the thigh. I then circumscribe the lower end of the tarsus and skin it to wel above the tibia joint. The leg is then skinned from the inside down to that junction and removed. Later, all loose bones are tied within the body cavity. When I am saving the entire skeleton of a large bird, I skin out both legs with the second technique described above.

If I am preparing a passerine schmoo and saving the entire skeleton, I do it slightly differently. After making my usual incision and loosening the skin on either side, I make a cut on each side with sharp scissors or a scalpel. The cut is at a right angle to the initial incision, from the incision to and down the inside of the leg to the lower end of the tarsus. The leg is then skinned without disarticulation. When the bird is sewn, one can catch each flap of each right-angle cut in a singl stitch in one's normal sewing pattern. The opening down the leg is ignored. As with a large bird, the tarsal skin is pulled posteriorly and tied to the stick in as normal a position as possible.

The pygostyle remains in the skin with the rectrices attached, and the body is then skinned forward. With a fat bird

the body may be kept in a plastic bag to prevent the feathers from touching it. The wings are disarticulated at their bases, and skinning proceeds to the base of the bill, which is detached from the body skin. After proper cleaning, the mouth is sewn shut. I use small stitches like those used on mammals.

If a measurable wing is to be left with the skin, the humerus is removed on that side and the meat is immediately removed from the radius and ulna by means of a standard external underwing incision. I try to leave the measurable wing on the same side of the bird as the complete tarsus. Once that wing is finished, the other is skinned to the terminal digit. It is important to remember that the base of the outermost primary is attached lengthwise to that bone and must be cut away from it. It is not unusual, even on a large bird, to lose that primary, and it is standard to lose several outer primaries on passerines, sometimes all of them on a cuckoo. Once that wing is finished, the entire skin is dampened and turned right side out (except, of course, for that wing). A large needle is threaded with button thread, used double, and several stitches are taken through the bases of the outermost primaries. These are dampened again and pushed right side out, and that wing is preened as neatly as possible. The needle is then pushed through the opening where the humerus was removed on the opposite side, and out between the radius and ulna. A stitch is made around the ulna a short distance from its distal end and the thread is pulled as snug as possible. Several more loops are taken, and the wings are

arranged. The wing that was entirely skinned out will never reshape perfectly and it will always be slightly 'klutzy'. Both wings may be skinned out completely if a complete skeleton is to be saved. In that case, the bases of both sets of primaries are sewn together, slightly farther apart than if stripped ulnae were being tied. The skin is now ready to be stuffed.

The eyes are standard, but they can be larger than normal to help replace the missing skull. A short, sharpened dowel is wrapped and put into the leg with the complete tarsus and is embedded in the joint. A short, blunt dowel is wrapped for the other leg.

I use either of two body types. I sometimes make a wadded paper body, about the size of that removed and bound with heavy thread, and thrust a sharpened 1/4" dowel through it with sufficient wood projecting for the neck. The neck is wrapped in cotton, with an enlarged ball at the head end covering the sharp point. Then, the entire body is lightly coated with cotton and the one-piece body is put into the skin. Before sewing, additional cotton can be added to the neck or to the flanks and lower abdomen, if needed.

One can also make an all-cotton body, using coarse brown packing cotton. A 1/4" dowel is wrapped with cotton for a length equal to that of the body, with a larger knob at one end to partly replace the skull. At the other end, 5"-6" (depending on the size of the bird) is left free of cotton, to provide a place to which to tie the legs after the skin is closed. The wrapped

dowel is put into place, working the knob into place between the eyes. Then a roll of heavier cotton is made, one-half the diameter of the removed body and of sufficient length that, when broken in half, it will form the two sides of the body. These are put into place. Finally, a large triangular piece of cotton is inserted, pushed forward to the mouth, and spread so that it covers the entire ventral aspect of the bird. This is tucked in, extra material is removed or added as necessary, and the body is sewn shut. This is essentially the Sutton method of body construction.

The legs are tied to the stick with the empty tarsus under the complete one. The wings and tail are adjusted and the specimen is braced for drying. I use scrap floor boards for dividers, so I can line up several large birds in a row for drying.

Needless to say, the skeleton and skin should be cross-referenced so that the skeleton can later be identified, if desired, to subspecies.

A specimen drying and carrying case for field use

Henri Ouellet

National Museum of Natural Sciences

P.O. Box 3443, Station D

Ottawa, Ontario, Canada

K1P 6P4

In the early 1970s after a few seasons of field work in central and northern Canada where climatic conditions had been particularly cool and humid it was felt necessary to improve upon the methods used in the field for drying and transporting freshly prepared specimens. After having examined all the available field material at the National Museum of Natural Sciences, it was concluded that the type of drying case used in the mammalogy section could serve as a model but that it was not entirely satisfactory for our purposes. With the assistance of the technical services of the museum a new concept was developed on the basis of the following criteria: light weight, portability, flexibility, good ventilation, large capacity, durability, good resistance to shocks, and imperviousness to dampness, insects and dust.

A new case incorporating all these features was thus designed and appeared to meet most requirements of field conditions. A few prototypes were produced but only two models were constructed with appropriate improvements: a small one for smaller specimens or for short periods in the field and a larger one for large specimens or for long field periods.

These cases are made of heavy gauge aluminium sheets welded and reinforced at the corners and at the rims to increase rigidity and sustain impacts. They have a fully removable cover and underside and are equipped with fine mesh fibreglass screens to prevent insects or other pests to enter when they are in the open position. The top is furnished with a lockable hasp in front whereas the back is retained by two interlocking folded reinforced ridges, one on the back of the cover and the other on the back of the case. The bottom cover is held in place by a number of spring-loaded retainers or pressure fasteners that can be sealed with waterproof tape for added security. The specimens, either for drying or shipping, are placed in drawers with a bottom of galvanized metal 1/2" (26 mm) mesh fastened to a wooden frame. The height of the frame of these trays varies but it was found that four sizes for the small case and five for the large one could accommodate all our requirements for birds ranging in size from a hummingbird to a duck or a grouse.

When the top cover and the bottom are removed the case can be properly supported to allow for a good circulation of air at the base so that freshly prepared specimens can be dried in less than 48 hours. However, the process can be accelerated when the case is used in combination with a catalytic space heater placed under it. The case needs then to be supported on three sides by boards or other cases. In this fashion a flow of dry warm air is circulated through the fresh specimens which can usually be dried overnight even in cool wet environment.

This type of case is now regularly used and has been utilized successfully under a variety of difficult climatic conditions. It proved to be very satisfactory and contributes significantly to the preparation of better specimens and their safe shipping.

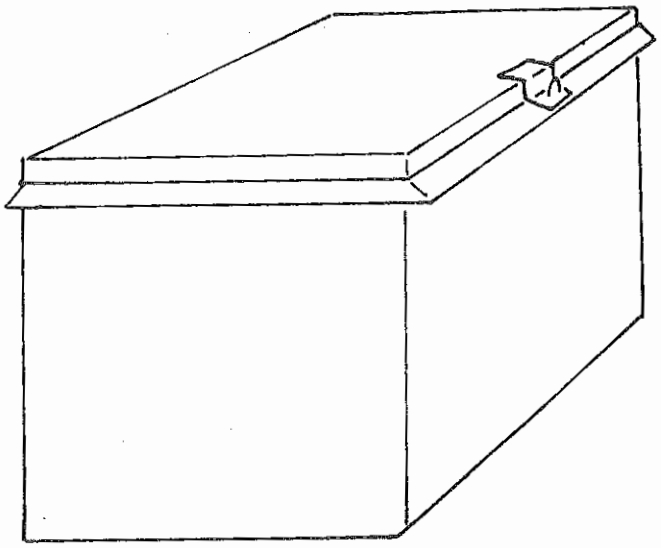
MEASUREMENTS

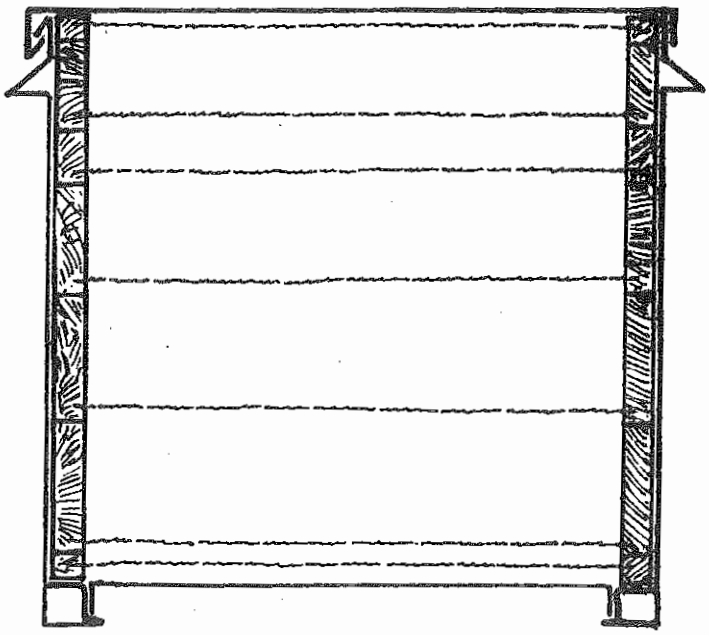
CASES

	Small	Large
Length	24" (62 cm)	28" (71 cm)
Depth	14" (36 cm)	18" (46 cm)
Height	14" (37 cm)	18" (46 cm)

TRAYS

Length	23 3/4" (61 cm)	27 5/8" (70 cm)
Depth	13 7/8" (35 cm)	17 5/8" (45 cm)
Height	1 1/2" (4 cm)	1 1/2" (4 cm)
	2" (5 cm)	2" (5 cm)
	3" (7.5 cm)	3" (5 cm)
	4" (10 cm)	4" (10 cm)
	----------	5" (15 cm)





SUMMARY OF TECHNIQUES TO PRESERVE FROZEN TISSUES

AMERICAN ORNITHOLOGISTS' UNION ANNUAL MEETING
UNIVERSITY OF PITTSBURGH, 7 AUGUST 1989
SHANNON J. HACKETT AND ROBERT M. ZINK
MUSEUM OF NATURAL SCIENCE
LOUISIANA STATE UNIVERSITY
BATON ROUGE, LA 70803
(594) 388-2855

WHY SAVE TISSUES?

Since the development of molecular biological techniques, frozen tissues have been a major resource for studies in evolutionary biology, systematics, genetics, biochemistry, and immunology. The interest in sampling genetic variation/differentiation, through analyses of proteins or nucleic acids, has resulted in an ever-growing resource base of frozen tissues of all sorts of plants and animals, dispersed throughout the research community. With ever-limiting opportunities for collection of specimens, it becomes important to maximize the materials preserved from each specimen; we feel that tissue should be saved from most organisms that are collected, especially those that are rare, endangered, or found in remote or disappearing habitats. Furthermore, several museums have made a commitment to preserve frozen tissues to accompany the more traditional skin, skeleton, and alcoholic collections. We hope to encourage field collectors and museum workers to preserve tissues as they do more traditional preparations, or to deposit tissues in a formally recognized tissue collection (see Dessauer et al. 1988).

The LSUMNS has the one of the largest tissue collections, housing tissue samples of fishes, amphibians, reptiles, mammals, and birds. We have approximately 14,000 bird specimens, which are available to qualified researchers upon entering into an exchange agreement. We currently have some funds for shipment of "orphaned" collections of frozen tissues to LSU, and will accept most donations. These tissues will be installed into our frozen tissue collection, curated by museum staff, and made available in accordance with our established grant policy.

HOW TO SAVE TISSUES

Probably the most important aspect of saving tissues is to preserve tissues at low temperatures as quickly as possible after death. The question that always arises is how long after death these are tissues usable. The answer to this question is "it depends." It depends on the ambient temperature (if > 26 ^{o}C freezing must be more rapid; Johnson et al. 1984). It also depends on the type of analyses one wishes to perform with the tissues. The extent of denaturation over time is different for different macromolecules and organelles; for example, proteins seem hardier than mitochondria. In general, DNA is very stable (see Houde and Braun 1988, Auk 105: 773-776), but for isolation of intact mitochondria and analysis of purified mitochondrial DNA (mtDNA), we suggest getting tissues into liquid nitrogen or dry ice (or into a buffer) within 1/2 to 1 hour. Any longer lowers yield of mtDNA. Other techniques allow study of mtDNA from tissues frozen in a few hours. For protein analyses, we recommend removing tissues and placing in liquid nitrogen or dry ice within 2 hours of collection. This may be inconvenient for field collectors, but we feel that it is never a bad idea to get the tissues frozen as fast as possible (see Johnson et al. [1984] for methods of rapidly removing samples from a series of specimens).

No one knows precisely how long proteins or nucleic acids remain physiologically active under field conditions. Protein stability under field conditions has been demonstrated for very few species. Moore and Yates (1983, J. Wildl. Manage. 47: 1166-1169) studied protein denaturation in 4 mammal species. They found that 95% of the proteins they examined were physiologically active in unfrozen tissues for at least 12 hours following death. Thus, it seems

that certain proteins are stable to degradation for long periods of time at 25 ^{o}C. Our personal experience would modify this conclusion. Not all proteins are stable to denaturation under those conditions, and for electrophoretic surveys requiring many loci, we recommend freezing as outlined above.

THINGS THAT YOU WILL NEED IN THE FIELD

dissecting equipment, scales, catalogues (personal and special one for frozen tissues)

storage vials that can go into liquid nitrogen or dry ice (depending on what you have)

supply of permanent marker pens

extra-heavy-duty aluminum foil wrap (in case something happens to the storage vials)

liquid nitrogen tank filled with liquid nitrogen or dry ice

1. STORAGE VIALS

We use two types of storage vials

(1) Nunc ™ tubes

expensive, used for liquid nitrogen

(2) Sarstedt ™ vials

less expensive, used for ultracold freezer or dry ice only

These sealable tubes are preferable to wrapping in aluminum foil, which can result in freezer burn to tissues when put in liquid nitrogen. If aluminum foil is your only option, use extra-heavy duty freezer foil. Try to transfer foil-wrapped tissue to storage tubes at a later date. Also, Nunc tubes have a shelf-life. Tubes that are older than 1.5 years will be brittle and shatter when placed in liquid nitrogen. Older tubes, however, can still be used for tissue storage in an ultracold freezer (-70 ^{o}C). Nunc tubes also come in various sizes. We recommend the larger tubes (4 ml or greater) for any bird larger than a Dendroica warbler.

2. LABELING

Use only permanent ink pens--ultra fine-point permanent pilot pens (SC-UF) or permanent marker sharpie pens. Do <u>not</u> label tubes with a rapidograph. This only etches the labeling information into the tube, and the label will rub off and become illegible. Label vials before tissue is placed in them (you won't be able to write on a wet cold tube). Label vials neatly with the following: personal catalogue number, scientific name, types of tissue in tube (L, H, K, and M for liver, heart, kidney, and muscle, respectively), and tissue number (if applicable). Try to write either the personal catalogue number or tissue number on the vial more than once.

Fill out the tissue catalogue, if applicable (see #4 below). Under remarks section, note such things as time elapsed before tissue was removed from bird and frozen, how long bird remained in freezer (and what type of freezer--regular or ultracold), and any other comments regarding state of tissue.

3. TAKING TISSUE

Our goal at the LSUMNS is to have at least 5 tissue samples of all taxa from each collecting locality. When "tissuing" a bird, take as much as possible. Fill the vial up to the fill line. Vials may rupture if filled past the fill line. For larger birds, take more than one tube of tissue. Label vials with appropriate tissue type, and also label how many tubes you have used (for example, 1 of 3, 2 of 3, 3 of 3). We tend to place tissue in tubes liver first, heart next, muscle on top, although this is only a suggestion.

It is important for some protein work to have samples of heart, liver, and muscle because certain isozymes show tissue specific expression. Kidney is useful and should be saved on most birds, especially small birds with not much liver. Other tissues--brain, testes, ovaries, blood, and feather pulp--have more limited use and are usually only saved for specific research projects. Testes and developing eggs can be a rich source of mitochondrial DNA. Sampling feather pulp is an important non-destructive means of sampling (see Mardsen and May 1984, Auk 101: 173-175; Haig and Oring 1988, Auk 105:260-267). Blood can also be taken from birds with only minor injury to the animal. Muscle biopsies are also useful (Baker 1981; Auk 98: 392-393).

4. TISSUE CATALOGUE

Each tissue specimen is entered into a tissue catalogue in the field. At LSU, our tissue catalogue contains the following information: tissue number, personal catalogue number, scientific name of specimen, locality information, and a remarks section. We assign each tissue sample its own unique number, similar to the catalogue number any specimen would be given upon curation. This number is written into the tissue catalogue, on the "voucher" specimen label, and into the personal catalogue. In the field, we carry a field tissue catalogue, and each collector or field party is assigned a block of tissue catalogue numbers. In this way, we can cross reference tissue samples with the personal catalogue number and a tissue catalogue. The tissue catalogue numbers, besides acting as a cross-referencing device making curation easier, also identify tissues from specific expeditions; for example, B9000-9800 may signify Peru 1986.

5. LIQUID NITROGEN

Liquid nitrogen (-196 ^{o}C) is the best choice for long-term field storage of tissues. For short-term storage, dry ice (-76 ^{o}C) and a thick-walled cooler will suffice. There are a number of liquid nitrogen refrigerators (dewars) available for field usage. These tanks can be obtained from the Cryogenic Equipment Department of Union Carbide Corporation. Sizes of these tanks range from those that contain 50 litres when full (lasting several months) to 10.4 litres (lasting 60 days). In addition, there are also dry shippers available. Dry shippers have an adsorbent, and no liquid nitrogen will spill. It might be possible to borrowing tanks from institutions that are involved in saving frozen tissues. Liquid nitrogen is available commercially in few towns and cities. If you will be in the field for an extended period, identify reliable sources of liquid nitrogen before you go. Universities, hospitals, welding companies, and mining and agricultural operations often carry liquid nitrogen or dry ice.

See Johnson et al. (1984, Wilson Bulletin 96: 543-560) for instructions regarding transporting liquid nitrogen tanks on airplanes and in foreign countries. Collectors at the Philadelphia Academy of Natural Sciences recommend filling extra Nunc tubes with water and placing in the liquid nitrogen tank when transporting tank. Liquid nitrogen often cannot be transported on airplanes, and must be poured out of the tank; these ice-filled Nunc tubes help keep tissues frozen during transport.

RECENT DEVELOPMENTS IN MOLECULAR BIOLOGY

Recent developments in the area of molecular biology are relevant to frozen tissues and molecular systematics. Demonstration of the long-term stability of nucleic acids (see Houde and Braun 1988 Auk 105: 773-776) provides a new and potentially exciting use for existing collections (skin, skeleton, alcoholic, as well as tissue). In 1985, Pääbo (Nature 314: 644-645) cloned and sequenced fragments of repetitive DNA from an ancient Egyptian mummy. In 1988, Pääbo et al. (Nucleic Acids Res. 16: 9975-9987) used the Polymerase Chain Reaction (PCR) to amplify and sequence nucleic acids from a 7000 year old human brain preserved in Florida. PCR allows for the amplification of copies of nucleic acids from a single copy of a gene, even from moderately degraded DNA. Higuchi et al. (1984, Nature 312: 282-284; 1987, J. Mol. Evol. 25: 283-287) isolated and sequenced DNA from the hide of an extinct equid, the quagga, and then compared these sequences to those of extant members of the horse family. Scientists have also been able to isolate DNA from chemically preserved tissues; Goelz et al. (1985, Biochem. Biophys. Res. Communications 130: 118-125) isolated DNA from human tissues that had been fixed in formaldehyde, preserved in alcohol, and then embedded in paraffin. Although DNA can be extracted and amplified from study skins, traditional collections are not the best source of macromolecules. At this time, we advocate the preservation of frozen tissues to maximize the yield of information from a specimen.

SELECTED REFERENCES FOR THE MAINTENANCE OF FROZEN TISSUE COLLECTIONS

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COMBINATION SPECIMENS A LA BURKE MUSEUM

Carol Spaw, Burke Museum DB-10
University of Washington, Seattle, WA 98195

Currently at the Burke Museum, more than 80% of incoming birds are prepared as combination specimens. These combinations include 1) round skin / bony spread wings, 2) round skin / skeletons, 3) flat skin / skeletons, and 4) skeleton / bony spread wings. Round skin and flat skin / skeleton combinations may sometimes include a boneless spread wing taken from the wing allocated to the skeleton or occasionally, a bony spread wing. While such specimens may not be as aesthetically pleasing as the traditional study skin, the greater amount of information preserved per bird makes these preparations more valuable for many types of research. The following sections elaborate on our methods of preparation.

1. ROUND SKIN / BONY SPREAD WINGS

We now prepare most of our skins with associated spread wings which display important plumage features that folded wings on skins cannot show. The Button-Stick method (details below) has minimized the extra time it takes to prepare a skin with one wing removed as well as the resulting asymmetry in the specimen. Indeed, this technique is so successful that we usually cannot distinguish skins having one or two wings without seeing their backs.

1a. *Detaching wing from specimen:* Begin the cut at the leading edge of the wing skin, near its attachment to the torso and distal to the scapulars which should remain with the skin part of the specimen. Cut through the humerus or disarticulate it at the elbow. The more perpendicular the cut is made to the bone, the smaller will be the resulting wing hole in the skin. This will later facilitate securing the button-stick in the skin.

1b. *Clean-out:* Remove the humerus and any muscles or fat near the cut. If there is little meat between the ulna and radius (as is the case in most passerines and small birds) , then the wing is ready to pin out. In larger birds, the muscles should be removed. Do not strip the secondaries from the ulna in

order to remove the muscles because it is difficult to rearrange them. Rather, remove the muscles through an incision made on the underside of the wing between the ulna and the radius, and then sew up the hole.

1c. *Label:* We tie labels through the propatagium, inserting one string of the label from the dorsal to the ventral side and tying the knot on the ventral side of the wing such that it lies near the attachment of the other string to the tag. This assures that as the label is moved, the knot will not be forced back and forth through the skin. When the knot is tied, a nice loop should be left such that the tie does not distort the patagium by synching feathers together. (see diagram)

1d. *Pin-out:* As a standard, wings are fully extended to show all flight feathers as well as primary slotting, if it's present. The placement of 4 pins accomplishes this. Lay the wing dorsal side up on the pinning board. Insert two pins in the propatagium, one through the wrist and one through the corner of the skin where the wing was initially cut, stretching the anterior edge of the wing taut. Insert the third pin through the skin at the elbow, angling the pin down to hold the secondaries down. Finally, with the fourth pin, extend the outermost primary until all of the primaries including slots are displayed. With additional pins, evenly space other primaries and secondaries. Usually this is necessary only on large birds or specimens that are partly dried from being frozen for too long. We leave wings pinned-out until there is no flexibility in the wrist joint; this may take weeks in very large birds! If unpinned too soon, the propatagium will shrink, causing the leading edge of the wing to wrinkle and the primaries to fold in. (see diagram)

1e. *Preparing round skin with only one wing:* The problem with preparing a one-winged study skin comes with tying the wings together. To solve this, Gary Shugart has developed the Button-Stick. Instead of tying the two ulnae together, tie the ulna left with the skin to a short stick. The distance between the ulna and button-stick should be the distance you'd normally leave between the ulnae or slightly longer. Then button the tied stick through the hole left by the detached wing. The stick should be long enough to keep from slipping back through the hole but short enough to remain hidden by scapulars and flank

feathers. As a rule of thumb, make the stick the same size as the ulna. Now the skin is ready to stuff as usual.

ROUND-SKIN / SKELETON (SPREAD WING) SPECIMENS

With round-skin / skeleton specimens, the entire skull (including the beak) and the bones from one wing and one leg are preserved with the skeleton. The pygostyle as well as the bones from the other wing and leg remain with the skin. The spread wing, if prepared, is usually boneless. The challenge for the preparator is to put things back in such a way that the plumage is displayed correctly and the stuffed skin resembles the original bird in spite of its missing parts.

There are two modifications of regular stuffing methods which apply to round-skin / skeletons. First, the supporting dowel, which is usually contained within the specimen, extends out through the mouth replacing the beak, as well as through the cloaca to support the remaining leg. Having this "peg" for a beak makes it possible to more accurately reconstruct the face. Second, the skull-less head skin is filled out with cotton.

2a. First choose which wing and leg will be kept with the skeleton. If some of the appendages are broken, keep both an intact wing and leg with the skeleton. When skinning out the bird, remember to disarticulate bones to be kept with the skeleton. Take any bill measurements before preparing the bird since the resulting skin will be beakless.

Skin-out / Skel-out: Skin out the specimen through either a lateral cut (see Flat Skin-Skeleton Section for details) or a mid-ventral cut. A lateral cut allows you to remove the skeleton in one piece and is usually faster, but the skin is more difficult to stuff symmetrically. When the only intact wing and leg are on opposite sides of the bird, both can more easily be kept with the skeleton when the specimen is skinned out through a mid-ventral incision. The directions which follow apply to a mid-ventral cut.

2b. Make the initial skin cuts. Cut the skin around the skeleton-side wing, close to its connection to the torso, leaving the scapulars with the skin. Disarticulate the elbow, removing the wing. Then make a mid-ventral incision, extending from the posterior area of the sternum down through the cloaca.

2c. Start skinning out the bird as you would a regular skin. On the skin side, disarticulate the knee or cut just below the knee, clean the tibiotarsus, and wrap it with cotton. Keep the intact femur from the skin side with the skeleton. On the skel side, skin the leg as far as it is feathered. Cut the skin there to free the leg, then pull the foot through the hole. Cut the skin along the leg to enlarge the leg hole if necessary. Cut through the vertebral column just above the pygostyle. (Check for bursa first.) Skin forward, up the back and breast of the bird, disarticulating the wings from the skeleton at the shoulders or elbows, and continue over the head to the beak.

2d. Where the skin meets the beak, cut the specimen's skin from the carcass. Leaving some of the rhamphotheca with the skin is OK, but do not leave any plumage with the carcass. You needn't make an extra cut to skin the heads of large-skulled birds such as waterfowl. Rather, skin the bird as far as possible and disarticulate the neck near the back of the head. Then from the outside cut the skin around the mouth as described above. Skin the head from the mouth back toward the neck, and remove the skull.

2e. Go back to the wings. Clean and tie the skin-side wing with a Button-Stick . Pull the feathers from the skeleton-side wing or, to preserve a boneless spread wing, skin out the wing, stripping the remiges from the ulna and manus.

Stuffing the skin:

2f. Secure the wing using the Button-Stick method. Insert cotton eyes into the skin.

2g. Cut a dowel long enough to extend beyond the mouth opening a short distance (the skin from the upper and lower mandibles will be tacked together around the dowel on both sides) and beyond the cloaca far enough to attach the leg to it. A short distance from one end of the dowel form a cotton skull by

feeding long, thin layers of cotton successively onto the twisting dowel (thanks to LSU for the cotton brain idea). The new "skull" should be small enough to feed up through the neck of the specimen and will partially fill out the head skin. (For long, narrow-necked species, see notes below.) Feed the wrapped end up through the neck, lodging the cotton skull in the head.

2h. Stuff the head and torso. Fill out the crown and sides of the head by stuffing small pieces of cotton into those areas through the mouth. Fill in the back. Insert a flat piece of cotton under the dowel to fill in the space between the dowel and the dorsal skin. (I often insert an extra piece of cotton along the back on the side with the missing wing, if the removed wing is especially large.) Make a body of cotton with a narrow twisted cotton "neck" at one end. Feed the "neck" of the body into the empty body skin through the ventral opening, then up the neck of the skin and out through the mouth hole. Holding the neck cotton with one hand, gently pull the torso skin down and over the cotton body with the other hand. Then adjust the length of the specimen's neck by pulling the neck cotton out further, if necessary, through the mouth, as you would in a regular skin specimen. Cut off the extra neck cotton, leaving sufficient cotton to fill out the throat and chin.

Birds with long and narrow necks are "stuffed" differently. Wrap the head, neck, and body portions of the dowel with layers of cotton until it is of sufficient diameter to fill out the neck. Insert the dowel up through the neck and out through the mouth as above. Often the cotton wrapping gets hung up on the skin. Dusting the inner neck skin with sawdust and/or wrapping thread around the cotton neck prior to feeding the dowel through the neck will help prevent this. Stuff the head skin and tack together the mandibles as above. Insert a neckless cotton body to fill the torso area and sew up the ventral cut.

2i. Sew together the mouth and ventral incisions. Using the protruding dowel as the bill substitute, tack the upper mandible skin to the lower mandible skin on each side, respectively, lining them up as they would orient around the actual beak were it there. Use as many separate stitches as is necessary to do this properly. Then sew together the ventral incision as in a regular skin.

2j. Tie the skin data tag to the leg and the leg to the portion of the dowel that extends though the cloaca.

2k. Complete the specimen. Pin or wrap the stuffed skin as you would a regular skin. Spread the tail and pin. If it seems that the skull-less head might mash flat while drying, prop the extended beak dowel up so that the head clears the pinning board. Pin out small boneless wings as directed in 1d. For birds with large wings, replace the ulna with a dowel before pinning the wing out. Boneless wings tend to be broader and shorter than bony wings; you must correct for this when pinning. (Note: We no longer routinely prepare boneless spread wings because they are weaker specimens and it is difficult to retain the original shape and feather arrangement.)

3. FLAT-SKIN / SKELETON / (SPREAD WING) SPECIMENS

Flat skin-skeleton specimens preserve all of the features of the stuffed skin-skeleton. In addition, since the skins are pinned out flat, any molt can be directly observed from the skin side, and pattern areas and colors are easier to measure. Furthermore, since the skins are not stuffed, they are faster to prepare and take up less storage space. Flats are skinned out through a single incision which extends the entire length of the bird. We make the incision laterally rather than ventrally so that the entire breast and back plumages may be viewed as complete units in the resulting specimen. As with the skin-skeleton preparation, the bird's bill, one wing, and one leg are kept with the skeleton, and the pygostyle and remaining appendages stay with the skin. However, because the specimen faces toward the right or the left depending on which side the lateral cut is made, appendages from the same side must remain with the skin, hence the skeleton.

3a. *Skin-out/ Skel-out:* The lateral cut is made along the side of the bird designated to "go with the skeleton". A complete cut from beak to cloaca may be made (a) all at once, initially, or (b) in two stages, skinning out the specimen between (more details below). I prefer the first plan for large, densely feathered birds and the latter for smaller birds. With either plan, begin on the skeleton side by cutting the skin completely around the base of the wing

leaving the scapulars with the skin. Then continue to cut from under the wing to the cloaca, cutting between the feather tracts of the breast and back and behind the leg. Cut the skin along the back side of the leg as far as it is feathered; skin the leg, freeing it by cutting around the scaled part of the leg.

Plan a: Extend this lateral cut forward to the beak. Position the specimen so that the neck and beak extend straight forward in line with the side of the torso that has been already cut. Begin your incision at the anterior side of the wing cut. Continue straight up the side of the neck, forward above the ear, and through the posterior corner of the eye. Continue to cut from the anterior corner of the eye, straight over to the edge of the beak. (Note: This cut can be made in the opposite direction.) Now, skin out the specimen, applying steps described in sections 2c-2d.

Plan b: Alternatively, the anterior part of the lateral cut can be made from the skin side rather than the feathered side of the neck and head, after the bird has been completely skinned out through the initial incision between the wing and cloaca. This way no feathers on the face or neck are cut. Skin the specimen out as in sections 2c-2d. After removing the skin from the skeleton, leave the neck and head skin turned inside out. In preparation for the neck / head cut, spread the torso out flat, feathered and wing side up. Then flatten the inverted neck and head skin, skel side up. The skel-side eye, facing up, should be more or less centered (see diagram). Starting where the dorsal and ventral feather tracts meet, cut up the neck, following its contour, forward to the head area and up over the ear to, then through the posterior corner of the eye. Finally, cut from the anterior corner of the eye to the mouth. To make a clean cut, use a very sharp blade and be careful to cut only the top layer of skin.

Pin-out:

3b. Lay the skin out flat. Check to be sure that the edges are not curled under. Arrange the feathers, smoothing them out and flattening them down with your fingernail. Tie a label through the propatagium as described in 1c.

3c. Position the skin for the pin-out. Place the skin from the lower mandible so that it is in line with the ventral neck skin. The tail will be pinned parallel to that chin / neck line. The eye and back of the head are situated to one side and up from the lower mandible. Spread the breast and back skin flat. Position the

closed wing at a slight angle, covering it with flank feathers. In passerines and small birds, turn the foot backwards and place it under the spread tail.

3d. Pin around the perimeter of the skin at its very edge rather than a feather tract. Catch the edge of the skin with the tip of the pin, pull out and pin down. Begin pinning at the head, pinning all of the points of skin from the mouth area and corners of the severed eyelid. Pin the skin from the back of the head, and the flap of skin from the side of the head. Pin out the breast skin, the back skin, the wing, and the spread tail (see diagram). Use as many pins as you need to pin all edges out flat. Unpinned areas, particularly in large, thick-skinned birds, will curl as they dry.

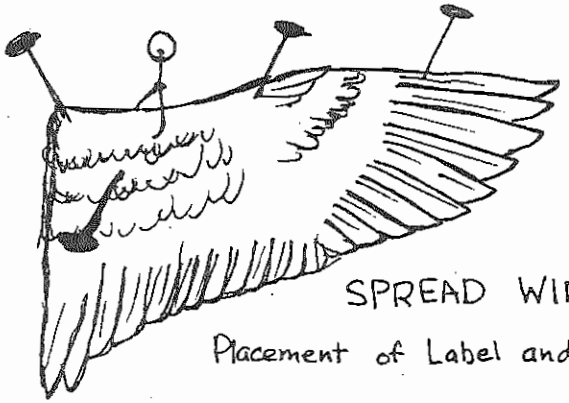
3e. Complete the specimen as in 2k.

4. SKELETON / BONY SPREAD WINGS

4a. Detach, clean out, label, and pin out the desired wing as described in 1a - 1d.

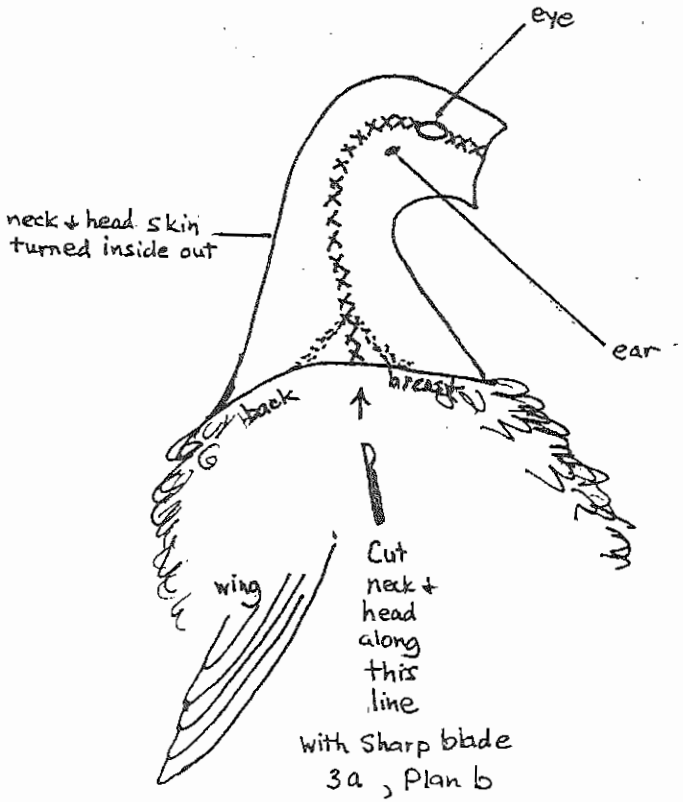
4b. Prepare the remaining carcass as a usual skeleton specimen.

ACKNOWLEDGEMENTS: My special thanks to the other Burke bird skinners including Chris Wood, Gary Shugart, Wendy Jackson, Dennis Paulson, and Sievert Rohwer, whose insights and comments are included in this paper.

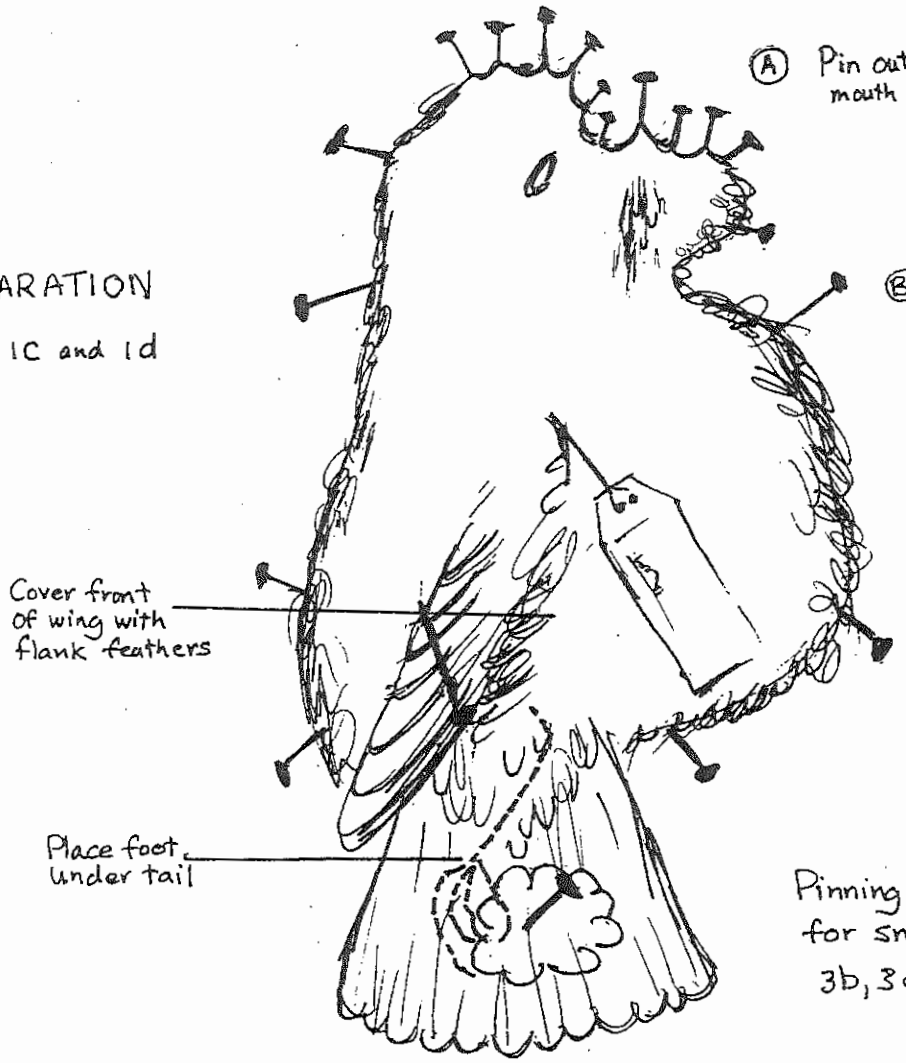


SPREAD WING PREPARATION

Placement of Label and Pins, 1c and 1d



With Sharp blade 3a, Plan b



Pinning arrangement for small birds 3b, 3c, 3d

FLAT SKIN PREPARATION

COLLECTION MANAGEMENT TECHNIQUES FOR FLAT SKINS AND OTHER NON-TRADITIONAL SKIN PREPARATIONS

Kimball L. Garrett
Collections Manager
Section of Ornithology
Natural History Museum of Los Angeles County

INTRODUCTION

Alternatives to standard "round" study skins of birds have long been employed in avian systematics studies. Flat skins and spread wings have been used in conjunction with particular research efforts (e.g. Clench 1970, Schreiber et al 1989), though they have become standard preparations in few collections. Cato (1986) noted only four collections housing flat skins and one housing spread wings. It is likely, however, that many additional such collections are likely to be identified by the survey currently being conducted by the Carnegie Museum of Natural History. With the waning of ornithology's rich tradition of saving aesthetically pleasing bird skins and throwing all the "good parts" away, an increased emphasis on anatomical material and alternative skinning methods can be expected. At present little written information on flat skin preparation and storage exists; preparation manuals such as Hangay and Dingley (1985), Anderson (1965) and Hall (1962) make no mention of avian flat skins. Knudsen (1972) mentions flat skins (in the sense of flattened, unstuffed study skins) only as a means of saving space in collections. In this paper I summarize some advantages and disadvantages of flat skins and other non-traditional skin preparations, outline preparation procedures, and make suggestions concerning storage, cataloging, and handling.

In most flat skin preparations the entire skin and associated plumage is removed, dried and stored flat. No special "tanning" procedures are employed. No skeletal elements are retained with the skin, therefore a complete skeleton (or a spirit-preserved body, minus skin) may be preserved. Flat skins allow thorough study of plumage, molt and pterylosis; the shape and pattern of spread wings may also be examined. In some collections spread wings are prepared from specimens that are otherwise skeletonized (wing skeletal elements may or may not be retained with the wing) or as an attached (or detached) adjunct to a traditional study skin.

Some disadvantages of flat skins may be identified. Standard external measurements must be recorded prior to preparation, and cannot be repeated. Because the skins must dry flat, field preparation (particularly of large skins) may be difficult. Even a well-prepared flat skin is an affront to the aesthetic sense of many ornithologists, although I argue that this should not be of concern. Finally, the long-term preservation prospects for flat skins, particularly those subjected to considerable handling, are poorly known. It is this concern that I would especially like to address.

PRESERVATION METHODOLOGIES

The flat skin technique employed at the Natural History Museum of Los Angeles County is a lineal descendent, via Glen Woolfenden, of the technique employed by Clench (1970) in her study of House Sparrow pterylosis. This technique differs from that suggested by Norris (1961), which was essentially a modified, unstuffed study skin affixed to a card with adhesives. Principles guiding the preparation of our flat skins are the following: (1) the entire skin, with associated plumage, is saved; (2) all skeletal elements are removed; (3) cutting across feather tracts is minimized; (4) natural wing shape is retained to the greatest extent possible; and (5) absorbants (corn meal, magnesium carbonate, etc.) are avoided or minimized (they obscure feather follicles).

A basic lateral cut is made from the gape to (and out) the tarsus; an extension of this cut extends from the lateral cut (at the base of the humerus) out the wing, following the wing bones. The opposing wing and leg are peeled off over the skeletal elements. The skin is freed at the base of the tail, and a cut is made lateral to the tail base to the vent to allow the skin to lie flat. [See diagram for clarification]. Muscle and connective tissue adhering to the skin is removed, and complete manual degreasing is attempted. The skin is washed if necessary. After final cleaning the skin is pinned out on a soft board. Interpin distance is small to maintain a smooth skin contour. When completely dry the skin is unpinned, sealed in a plastic bag, and frozen as a precaution against pests. Freezing is performed in a standard commercial chest freezer at -20 C for a minimum period of 48 hr.

In my experience, it takes about 50% longer to remove a skin for a flat preparation (versus a traditional round study skin), but time spent making up the skin (i.e. pinning it out) is about 50% less.

STORAGE METHODOLOGIES

Long-term storage of any vertebrate skin material (and in fact most museum specimens and artifacts) is governed by a series of basic requirements:

(1) an environment free of biological pests
(2) absence of light
(3) a cool, constant temperature
(4) a stable, moderate relative humidity
(5) non-reactive storage materials
(6) no preventive fumigation
(7) minimal physical stress in storage
(8) minimal handling stress

The first six requirements are addressed on the collection-wide

level and require adequate staffing, building and storage facilities, monitoring, housekeeping protocols, and use of stable storage materials. Requirements (7) and (8) demand solutions unique to flat skins. Recall that the flat skin specimen composite consists solely of proteinaceous material (the skin) and cellulosic material (the stringed tag). No cotton, wires, wood dowels, or other supports are used. Potential reactions with the environment and with storage materials are therefore reduced.

At the Natural History Museum of Los Angeles County we have addressed some of these problems, and have long-term plans to confront the others. Flat skins are stored in steel cases with steel drawers. Enamel paints used on these (Interior Steel) cases do out-gas, contributing to a more chemically reactive internal case environment. Perhaps a preferred solution is the use of non-reactive electrostatically applied polyester powder paints. Large flat skins (e.g. pelecaniform birds) are folded, placed in polyethylene bags, and stacked within drawers. We are now in the process of storing smaller flat skins by placing each skin (internal skin-side down) on a sheet of blotter paper, which in turn is placed in a polyethylene bag with an incomplete heat seal. The blotter stock should be of a moderately heavy weight (e.g. 30-50 weight) to provide support for the skin. It should have high stability, meaning lignin-free and pH-neutral qualities (beware simple descriptions such as "archival quality", "acid-free", or "conservation quality"). Acceptable blotter material is available from Archivart and other suppliers. We use polyethylene bags (supplied by Bradley) and a TEW TISH-300 impulse heat sealer. While complete seals are used for the freezing procedure, skins are stored in incompletely sealed bags to guard against moisture build-up within a small, closed micro-environment. Blotters can be replaced as grease deposition necessitates.

Storage of flat skins in polyethylene bags with a blotter support allows selection and examination of specimens without direct handling. It is simple to ensure that specimen tags are visible without removing the skin from the bag; a catalog number may also be written directly on the bag with a Sharpie or similar marking pen. Conspecific specimens may be stacked and housed within a labeled paper folder. One advantage of bagging is that loose feathers are retained with the specimen without having to be reattached.

LACM flat skins are stored in their own cases, separate from the study skin collection. Spread wings are stored with flat skins, unless they are detached from a study skin (in which case they are stored adjacent to the skin).

CATALOGING AND LABELING

The two most common methods of label attachment for flat skins are: (1) label strung through one of the eye holes; or (2) label sewn through the patagial region. At LACM we use the former method with considerable success; I have yet to encounter an instance where a label strung through an eye hole was lost or caused tearing in the skin. Labels strung through the patagium are standard for detached spread wings.

Flat skins at LACM are assigned a primary sequential catalog number, as are all other preparation natures. Skeletal or spirit material from the same individual specimen is covered by the same catalog number. While some collections maintain separate catalogs for skins and skeletons, there are clear advantages of a single numerical system where skin and anatomical material from the same individual receive the same catalog number. Specimen natures are coded in our database ("ORNCAT", using CURATOR software) as follows:

FS = flat skin only
FB = flat skin with complete skeleton
FA = flat skin with alcoholic carcass
WO = wing(s) only
SN = complete skeleton only
AL = alcoholic (entire)
SS = study skin (only)
KB = study skin with body skeleton
SA = study skin with alcoholic body
etc.

Computer searches based on the LACM catalog database can simply specify nature-of-specimen code(s) to obtain listings of skeletons, flat skins, study skins, or any other preparation type.

CONDITION ASSESSMENTS

Because flat skins (particularly those of passerines) are thin, they would seem to be susceptible to more mechanical damage from improper storage and routine handling than traditional round skins. I undertook a casual survey of the condition of 275 flat skins at LACM, including 120 passerines, 120 Pelecanus occidentalis, and 35 Uria. I looked for tears, grease damage, feather loss, label loss, and insect damage. As no labels were lost, I concluded that the eye holes, with their dried connective tissue serving as reinforcement, are suitable sites for label attachment. Insect damage was limited to several well-defined outbreaks, all of which could be traced to lapses in specimen installation, poor case construction, housekeeping, or monitoring procedures. Of primary concern was

the cigarette beetle (Lasioderma), although infestations in flat skin cases were no more frequent than in study skin cases.

Discoloration and slight feather loss from inadequate degreasing is a recurring problem, particularly among procellariiforms; grease damage was infrequent among the taxa examined in my assessment. Concerns about grease damage are not qualitatively different between flat skins and study skins; more extensive cutting in flat skins, however, exposes more feathered regions to wicking of fats and oils. Adequate degreasing of all skin preparations is essential.

Skin tears were found in no Uria and in only five of the 120 Pelecanus. All tears in pelican skins were on the neck, and perhaps resulted from excessive stretching combined with rapid drying during the preparation process. Tears did not coincide with the site where the neck was folded for storage. By contrast, 35 of the 120 passerine flat skins examined (29%) showed one or more tear. Tear locations were:

Base of tail (14)
Base of open wing (11)
Apteria bordering dorsal tract (7)
Base of open leg (5)
Head/neck (4)

Some of these tears undoubtedly occurred during preparation (drying or unpinning); others may have resulted from handling. The possibility of handling-induced damage led to the storage technique employing blotter boards and polyethylene bags.

SUMMARY

Flat skins and other non-traditional skin preparations are increasingly used as tools for systematic research. Few standards have been developed, however, to guide collection managers in the labeling, storage and handling of such preparations. While storage concerns for flat skins are similar to those of traditional "round" skins, additional efforts to minimize physical damage from storage and handling are suggested.

Any analysis of the advantages and disadvantages of flat skins (or any other preparation technique) can only be attempted with respect to the anticipated and potential research uses to which they may be subjected. Decisions must relegate individual specimens to the sometimes conflicting goals of consumptive analytical use and long-term preservation. Increased dialogue among preparators, curators, collections care professionals and researchers in making these decisions is of utmost importance.

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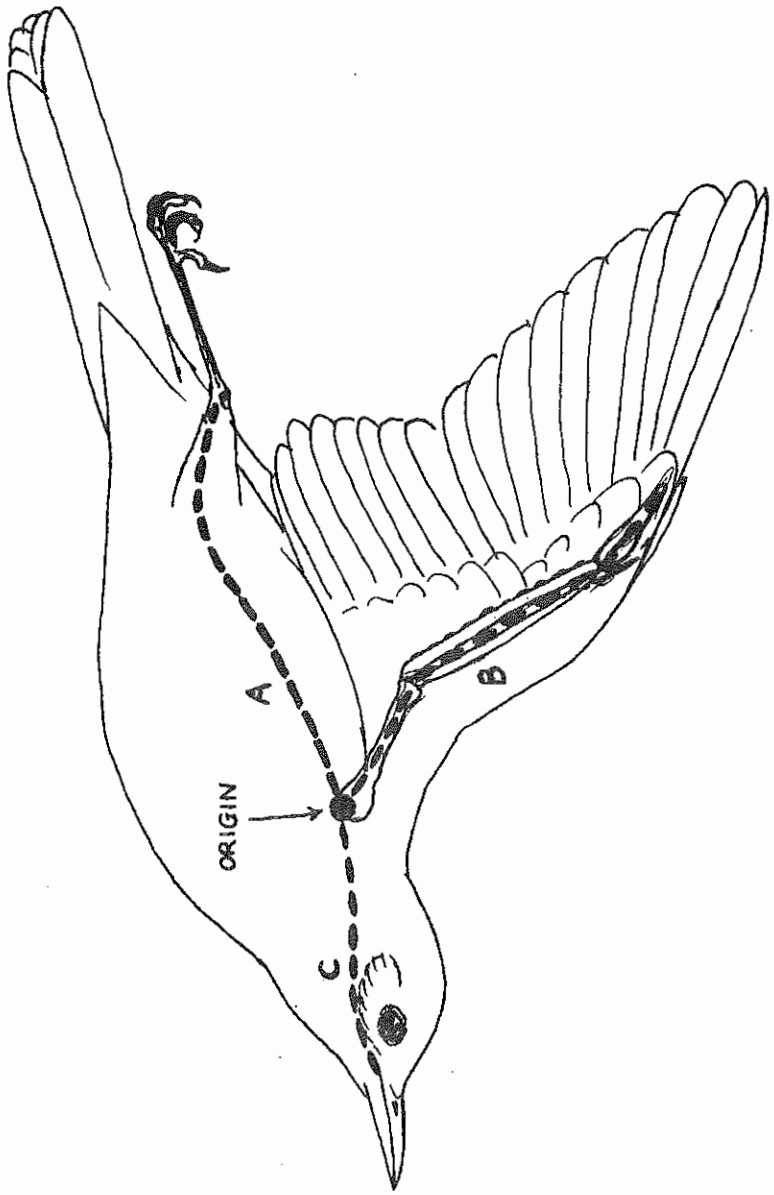
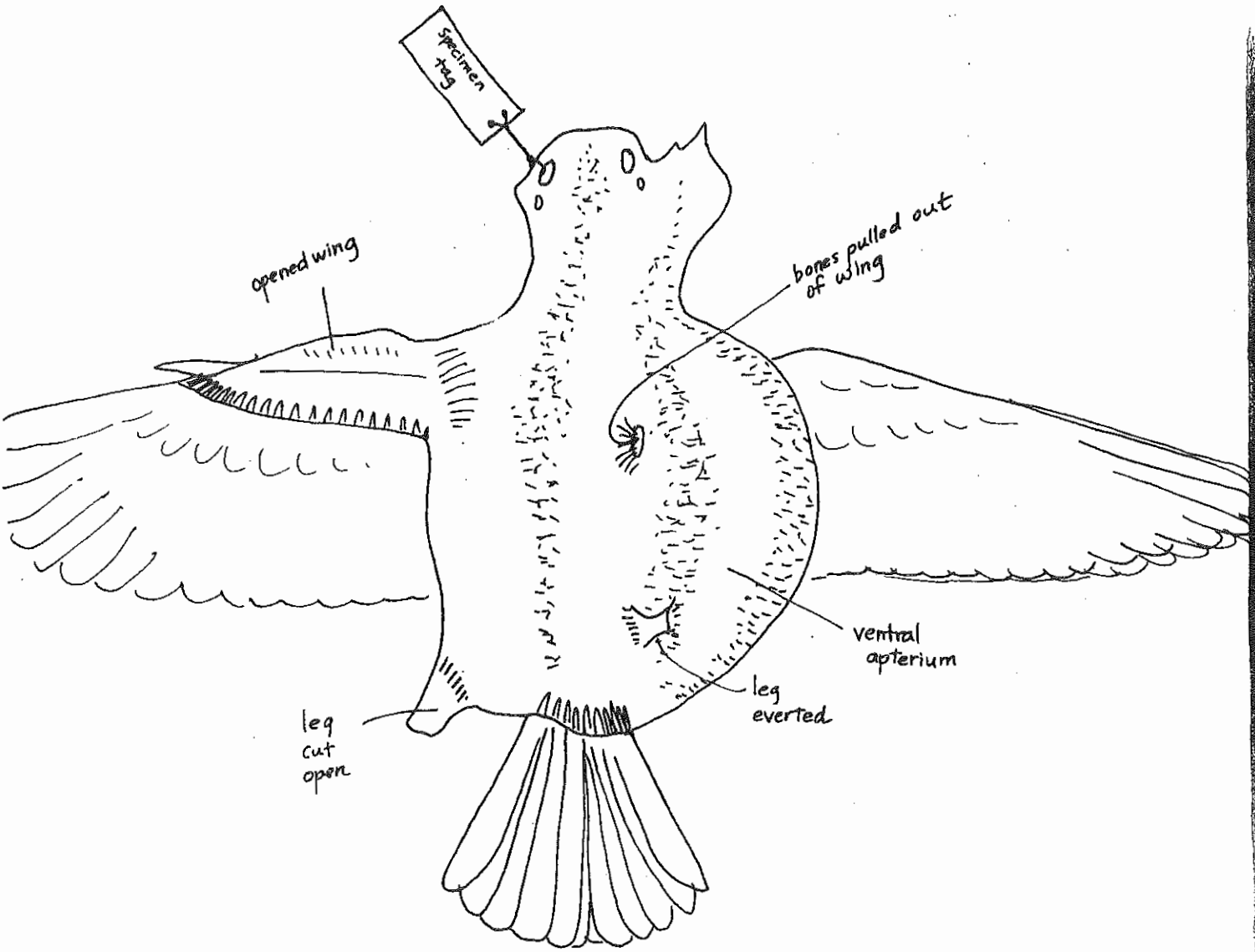


LACM flat skin technique: basic cuts



LACM Flat Skin – topography

Eremophila alpestris flat skin
photocopied 06 OCT 1988

NA: USA: CALIF; Los Angeles Co., Antelope Valley, vic. Lancaster (FB)
14 OCT. 1985
26.5 g; little fat
C12.3 W102 T21.4 Tail 63
Prep: SLM 801
Coll: K.L. Garrett

BIRD-SPECIMEN COLLECTION AND PREPARATION: A USER'S VIEW

Dennis R. Paulson
Burke Museum DB-10
University of Washington
Seattle, WA 98195

Rather than emphasizing a specific topic, I have chosen to present a personal overview of bird-specimen preparation, beginning with the collection of specimens. From working with specimens at many museums, I have been impressed by the inadequacies in bird collections that go far beyond the dos and don'ts of varied methods of preparation. Although most of these points are obvious to experienced preparators and collectors, and many have been made before (e.g., Van Tyne 1952, Stiles 1983, Johnson et al. 1984), I present them for further emphasis.

The discussion is organized around four basic principles: (1) what specimens to collect; (2) specimen data; (3) preparation types; and (4) preparation methods.

WHAT SPECIMENS TO COLLECT

This will be an idiosyncratic decision, based on the goals and objectives of both the museum and its individual staff members, and perhaps a certain amount of tradition. Objective considerations would dictate the collection of species that are most needed for ornithological research of all kinds, and these should include rare species, with special emphasis on salvaging specimens of endangered or threatened species if they become available; rarely collected species such as pelagic birds, swifts, rails and owls; very local species that can be best sampled by the local museum(s); and geographically variable species for which samples from all regions are desirable.

At some point in time there will be a summary of skins in all collections to parallel those of skeletons and preserved specimens that we now have. The ornithological community will then have a clear mandate of "target" species to be sampled, often manifested in the need to sample particular geographic regions, as pointed out by Jenkinson and Wood (1985).

Lists of study-skin holdings will not address the particular need for series of specimens in different *plumages*. Some species have been commonly sampled on their breeding but not their wintering grounds, or vice versa. Some bird species exhibit surprisingly many plumages, as detailed study of known-age specimens is revealing. Most immature birds that are widely dispersed—many seabirds, for example—are poorly sampled. At the Burke Museum, inventorying skins by plumage type has revealed the gaps in our collection clearly.

We badly need a guide to nonpasserine bird plumages along the lines of the passerine guide by Pyle et al. (1987). Even a list of all the plumages manifested in each bird species (still to be determined in some!) would be a great boon to collection

managers for specimen inventories. Computerized databases of collections should include plumage types for printouts to be of maximal value.

Molting specimens, often shunned in the past, should have high priority for preservation. Birds in molt may be retiring, are often south of the U.S. border (and thus of less interest to collectors than the tropical residents), occur during a relatively narrow time window, and are often rejected for study skins because they are scruffy. They are accordingly frustratingly rare in collections and should be high priority for collectors. I have been shocked at how many shorebirds I have had to examine in collections even to begin to understand their patterns of molt. Most of the relatively complete information on molt comes from field workers who have examined large series of birds in life.

SPECIMEN DATA

I have been surprised at how much potential information is absent from specimen tags, and this is not only a problem of past collecting. Probably the only way we will ever understand the plumage changes of birds is to have series of known-age specimens with which to work. This goes for passerines, for which skull ossification data on tags are not as common as one might think. Only certain collections and collectors have routinely recorded this variable, and few have quantified it (e.g., skull 25% ossified). On the other side of the coin, many collectors thought that nonpasserines could be aged in the same way, with the result that many juvenile shorebird skins are labelled "adult—skull ossified."

The vast majority of large-bird specimens lack information pertinent to assessing their age, and we need to examine many more seabirds and hawks, just to name two groups, to work out their plumage changes. Fortunately, in at least some groups the bursa of Fabricius is retained for several years. For example, bursa data from recent series of Tufted Puffins taken at sea finally allow an understanding of plumage changes in that species not possible from the examination of skins.

Although collectors have recorded gonad condition (size) for many years, much information about reproductive state is still lost every time a carcass is discarded. Both length and width of both testes should be measured, so testis *volume* can be roughly estimated. Many preparators have measured only the longer left testis, when in fact the shorter, wider right testis may be larger in volume. Enlargement of the vasa deferentia or oviduct should be noted. Questions about reproductive cycles may ultimately have to be answered by routinely preserving gonads from prepared birds.

Brood patches, not always easy to detect on a study skin, should be noted before preparation. Edematous brood patches indicate an incubating bird, while defeathered but dry patches occur from the young-feeding period to the fall molt. I have recently distinguished early southbound migrant shorebirds from summering

ones by the presence of brood patches. "No brood patches" is a worthwhile datum for a bird that the preparator feels might have had them.

Even such a thorough compendium as Dunning (1984) is inadequate for weight data on many North American birds. Series of different species were taken in different seasons, when weights can vary tremendously, and only gross comparisons among species are possible. Information about weight is still badly needed for most bird species. Weights on specimens must be fresh and must be accompanied by information on fat condition taken in a standard way. For museums that can afford it, there is now a machine that will determine the exact fat content of specimens in the 40-600 g range (Walsberg 1988).

Bare-part colors are not routinely recorded by many collectors because adequately known for most bird species. Many seabirds change bare-part colors with age, but this is neither apparent from the literature nor from examination of many specimen tags. I have discovered (originally from photographs) that the legs of many adult shorebirds change color seasonally. This is of interest for a variety of reasons but has been remarked for only one species (Wilson's Phalarope) in the literature. Similar color changes in other taxa, differences among closely related species, and, of course, colors of rarely collected species all need to be further documented.

It is critical that color notes be taken at the time of collection, as there is much postmortem change in at least some colors and some species. Color matching would be preferred, but even simple descriptions are of great value.

Studies of molt are tedious and would be greatly facilitated by descriptions taken during preparation. Midwing molt is simple to assess in a fresh bird but much more difficult in a skin. Saving an extended wing is an excellent alternative. Molt of contour feathers is easy to assess while the skin is turned inside out. Even more attractive is the reduction of wear and tear on skins—perhaps greatest during molt studies—provided by such information on the tag.

Data recording should be standardized among preparators: fat condition, molt condition, skull ossification, bursa measurements and way of recording colors (does "legs yellow" and "feet yellow" mean the same thing?).

PREPARATION TYPES

When specimens are collected, there are still decisions to be made about the type of preparation. These are typically apportioned among skins and skeletons. In a minority of museums, specimens are preserved in spirits, in fewer yet as tissue samples. I will confine my comments to study skins and extended wings.

Spirit specimens and skeletons are much rarer in collections than study skins, which points to the value of saving specimens as one of the first two preparation

types. Nevertheless, even with series of thousands of skins of some species available, they are still not adequate to answer all the questions that might be asked of them, especially because of incomplete data on many of them.

It is the substantial variation in bird plumages, even within species, that drives the need for large samples. Variation is by group (age, sex, population, morph) but also by individual. Thus the variation in each species must be understood well enough to determine which specimens are most valuable to preserve as skins (say, if competing with a need for skeletons or pickles).

Lists are now available to allow us to determine whether a bird is especially valuable as a skeleton or spirit specimen, and eventually there will be similar lists for skins. When we began our extended-wing collection at the Burke Museum, we had no idea whether such specimens existed in other collections, and this resource should be inventoried as well. Large series of wings should be preserved for species with patterned wings, and special attempt should be made to collect specimens during the period of wing molt.

Combination specimens (skin/skeletons, etc.) are the wave of the future, allowing maximal use of each bird. Although bird specimens can be equated to information in a data bank, each specimen is unique, unlike books in libraries. Therefore it will behoove us to maximize the information stored from every one.

PREPARATION METHODS

Some Burke Museum preparators feel that ulna stripping allows one to make better (by fixing the wings more anteriorly and dorsally) as well as quicker skins. I suppose this technique will remain controversial as well as individual. I have examined more skins of shorebirds than any other major taxon, and these birds, with their long tertials, suffer the most from sloppy ulna stripping. In some specimens, the tertials have slipped around and are between the wing and the skin!

Burke preparators have perfected ulna stripping, leaving the entire ulna with the skin and taking special care to rearrange the inner secondaries and tertials, but my attitude is still that the flight feathers lie most naturally on an unstripped ulna.

The distortion of the original pattern on a study skin can be a severe problem in birds with complex patterns. Color patterns are a result of individual pattern elements (fringes, tips, bars, etc.) on individual feathers, often associated with particular tracts, and on skins they often become illegible. The distinctive patterns on the backs of many shorebirds are a consequence of the scapular rows lining up, and they become disaligned on study skins. I have not yet discovered any way to prevent this, but it would be a worthwhile project. The best solution to date is individual feather alignment before careful wrapping and/or pinning.

In preparing combination skin/skeletons, which involves removing the bill from the skin, preparators must exercize great care to avoid distorting head patterns of such specimens.

Incisions made in study skins are usually ventral, occasionally lateral or dorsal, and there are advantages to each method. However, Colleen Nelson has called attention to the fact that downy young have complex patterns that can be strongly distorted by cutting anywhere but ventrally.

Extended-wing preparations are uncommon enough—routine in only a few collections—to be very exciting. I am in favor of removing such wings from their respective skins, as leaving them on the skin compromises storage of the specimen and will preclude against large series. No standards have been set for wing extension, and this is overdue. At the Burke Museum we have attempted standards, but it is equally obvious that some variation in method is inherently of value, as these specimens will have a variety of uses. Wing area can be measured at full extension, wing shape can be assessed over degrees of extension (soaring birds extend their wings more than flapping ones), and wing patterns can be analyzed over the same variation. Birds don't maintain their wings in a single configuration, so why should museums?

And this brings me to the statement that we are bound by traditional preparation methods that should be questioned just as vigorously as any other traditions. Crested birds are often prepared with heads turned so their crests are evident, but how often do we experiment with other methods? Preparation type could be idiosyncratic when appropriate or, better yet, variable to best express different features of the bird. For example, at least some individuals of birds that have complex belly patterns should be prepared with lateral or dorsal incisions. Gary Shugart has recently prepared series of "rightsideup" downy young to show their dorsal patterns more effectively.

Having worked out the techniques of remaking skins, at the Burke we are starting to augment our wing collection by removing and extending wings from skins of rare species. It is a rapid and effective way to round out this special collection and could readily be done with specimens of extinct species along with the removal of their skeletons as described by Olson et al. (1987).

APPENDIX: PUBLIC RELATIONS

With regard to how many specimens to collect, the one fact that is clear is that museums are not collecting enough! Part of this of course is due to the limited staff and budgets of most museums, but there is also constant pressure to reduce scientific collecting activities. Both the general public and permitting agencies are often critical of bird collecting, which needs to be put in perspective. Museum curators should analyze their collecting efforts and make a special point of relating them to bird populations.

For this purpose, I analyzed bird numbers in Washington, estimating from breeding-bird censuses in *American Birds* a nesting population of land birds averaging at least 200 pairs/km^2. Multiplying this by the state's area and assuming 2 surviving young/pair, I calculated 70 million birds produced each year. The Burke Museum collected about 700 birds during a "good" year, or 0.001% (*one out of every 100,000*) of the number produced.

Furthermore, from data published by Banks (1979), I calculated that about 20 birds per species were collected by scientific collectors during the early 1970s, and, if anything, this number is probably smaller now. Figures such as these should be on the lips of all museum ornithologists to make it clear that our efforts are inconsequential to bird populations.

The only critique that can appropriately be leveled toward museum curators is that in many cases specimens are still not being adequately used, from the standpoint of both data recorded and parts retained.

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The Use of Hexanes for Museum Specimen Preparation

Introduction

It's no secret how much damage a fatty, greasy specimen can do to other neighboring specimens when it starts oozing its contents.[1] Museum preparators have dealt with the problem for some time, and a number of tactics have been devised. The use of organic solvents is one of these, especially if one can be found that is both efficient and relatively easy to use. There are, of course, several considerations that immediately come to mind: 1) how might the solvent adversely affect the preparator using the substance and 2) how might the solvent adversely affect the specimen. If you are a preparator, the former consideration may take precedence. If, however, you are the curator in charge, you will no doubt consider the latter as most important.[2] At the N.C. State Museum we are currently using an organic solvent, hexanes, that when used cautiously, we think satisfies both concerns as well as others.

I will discuss hexanes in general, how they relate to what we're doing, and describe our setup and discuss results. We have been using this system for the last 10 years and could not have efficiently developed our marine bird collection without it.

Hexanes

The term "hexanes" refers to both the straight-chain hydrocarbon, C_6H_{14}, and branched hydrocarbons of the same formula (isohexanes). The straight-chain form is known as n-hexane. It is widely-used pure or as a commercial grade solvent for industrial purposes (see Uses). Commercial hexane is a narrow-boiling mixture of n-hexane, isohexanes (2-methyl-, 3-methylpenture, 2, 2-dimethyl-, 2, 3-dimethylbutane), and methylcyclopentane, cyclohexane and

benzene. n-Hexane is isolated from natural gas and crude oil. It is a small molecule[3] similar to white gas.[4]

NCSM Preparation Facilities

We[5] work with hexanes under a lab-type hood equipped with an exhaust fan (actual efficiency unknown). After skinning, feather tracts and any other areas with large amounts of fat need to be scraped. The hexanes will not dissolve large chunks of fat, nor do they work well when large sheets of membranous tissue are covering areas to be degreased. Before degreasing, specimens are washed in soapy cold-water.[6] As much water as possible is squeezed out after the rinse, before placing the specimen in a metal container, with hexanes in it (do not use plastic). We soak the specimens at room temperature (theoretically, a higher temperature would be even better - see Fats below). A 700 gram bird such as a Cory's Shearwater is placed in about 1 1/2 gallons of hexanes. Specimens are left in the hexanes from 2 hrs. to overnight depending on the bird and/or the timing.[7]

After soaking, specimens are again squeezed to remove as much hexanes as possible.[8] Specimens are then dried in a tumbler, using either sawdust (large birds) or cornmeal (small birds). Since the specimen may still contain a signficant amount of hexanes after squeezing, we blow-dry it with cold compressed air a few minutes under the hood to minimize exposure to the preparator. Because hexanes displace water and then evaporate quickly, the tumbling process should be monitored closely, to avoid damaging flight feathers.[9] A Cory's Shearwater dries nearly completely in 5 minutes in our tumbler. After tumbling, we blow-dry the specimen with cold compressed air to remove drying medium and to complete the drying process. Specimens tumbled in coarse sawdust do not dry as well as those in cornmeal or fine sawdust and require a little more blow-drying.

We reuse hexanes until the solution takes on a dull golden or yellow color. To re-use the solution it should be stored in a freezer for at least a day to allow the water to separate and the fats and debris to settle to the bottom. We then decant. We dispose of the spent solution by evaporation. Local agencies responsible for chemical disposal recommend evaporating one gallon at a time over cement on a hot day. Do not pour hexanes down sewers (flammability).[10] The recommendation for large-scale industrial disposal is incineration.

We buy our material from Fisher Scientific in 55 gallon drums, but it can be purchased in smaller lots of 4 liters or 20 liters. We estimate our hexanes cost to be about 0.30-0.50 cents per bird.

Uses

Commercial, or technical, hexanes are widely used for industrial purposes. The partial list provided might help in locating a local inexpensive source. Hexanes are used:

1. in motor and aviation fuel
2. as a solvent for extracting oils from seeds
3. as a solvent in the synthesis of polyolefins, synthetic rubber and other polymers
4. as a solvent in quick-drying rubber cements and certain two-solvent-system adhesives
5. in pharmaceutical industrial reactions

Physical properties

Hexanes are a very volatile, colorless liquid with a faint, gasoline-like odor.[11] The following information pertains to hexanes as purchased from Fisher Scientific, Chemical Division.

Boiling Point: 136F (58C)	Autoignition Temp: 437F (223C)
Melting Point: -139F (-95C)	Specific Gravity: 0.7
Flash Point: -7F (-22C)	Vapor Pressure: 124 mm Hg @ 20C
MW: 86.20	MF: C_6H_{14}

Hexanes are insoluble in water, but are miscible in alcohol, chloroform and ether.

Vapors are heavier than air and may travel a considerable distance to a source of ignition and flashback. Nevertheless they are not as volatile as white gas and we think hexanes are better at degreasing.[13]

Reactivity

Hexanes are stable up to the boiling point at normal pressures. They are incompatible with strong oxidizers (violent reactions). Thermal decomposition may release acrid smoke and irritating fumes.

Exposure limits and Health effects

500 ppm (1800 mg/m3) OSHA TWA (n-hexane)
50 ppm (180 mg/m3) ACGIH TWA (n-hexane)
500 ppm (1800 mg/m3) ACGIH TWA (other isomers of hexane)
1000 ppm (3600 mg/m3) ACGIH/STEL (other isomers of hexane)

Hexanes are a mild eye, mucous membrane and skin irritant. Acute exposure may cause narcosis and gastrointestinal irritation. Chronic inhalation may cause peripehral neuropathy;[14] chronic skin contact may cause dermatitis.

Inhalation: a level of 5000 ppm is immediately dangerous to life or health;[15] acute exposure may cause irritation of the upper repiratory tract, light-headedness, nausea, vomiting, narcosis and headache.[16] In humans, 2000 ppm for 10 minutes produced no effects, but there are reports of slight nausea, headache and irritations of the eyes and throat at 1400-1500 ppm.

Protective equipment

One should provide local exhaust or general dilution ventilation to meet published exposure limits. Ventilation equipment must be explosion-proof.[17]

One must wear appropriate protective gloves to prevent direct skin contact, as well as safety goggles. Contact lenses should not be worn.

Storage containers should be metal, not plastic. We use metal safety storage containers.

Fats

Most animal fats have >16 carbon atoms in fatty acid molecules, and marine animal fat has a high % of molecules with >20 C atoms; (rapeseed oil also has >16C, and hexane is used to extract this oil)

At temperatures above their melting points, fats and oils are freely miscible with most organic solvents except alcohols; at temps far below their mp's, fats are only very slightly soluble.

John Gerwin, Collection Manager
N.C. State Museum
Raleigh, NC 27611

(footnotes contributed by DSL)

[1]not to mention the original specimen itself and its tag

[2]if I ever get a good preparator I will be concerned with both

[3]species

[4]but with distinct vocalizations

[5]the "we" translates "I shoot 'em, John stuffs 'em"

[6]one can run the water through a fine mesh (nylon stocking toes are good) and contribute to your museum's insect and mite collection

[7]long-term soaking (longer than 24 hrs) on the skinned birds may cause slight twisting of body feathers

[8]wear rubber gloves

[9]In reality even if a bird is not fat and greasy but needs to be washed, it is worth putting it in hexanes to displace water. This way it is in the dryer for a shorter period and there is less damage to feather tips.

[10]Raleigh has more degreased rats than Chapel Hill

[11]the odor does not linger as it does in gasoline, however

[12]John has given up smoking and heated discussions

[13] also see Wood's (1988) discussion of hexane on bird feathers

[14]this is hard for us to evaluate because of high base-line neuropathy of NCSM collection managers

[15]sometimes

[16]only both if you get past 15

[17]we keep our specimens in another building

PREPARING BIRD SKINS FOR CLASSROOM USE

by

Stanley W. Harris
Department of Wildlife
Humboldt State University

The major problems with student use of bird skins are broken heads, tails and feet and lost labels. Breakage of wings and separation of support sticks from bodies are secondary problems. This breakage can be reduced by proper instruction of users in careful handling of skins. It is important that this instruction be continuous during use and be extended not only to student users, but also to laboratory assistants and faculty members. In spite of careful and constant admonitions to be careful, some skins are still occasionally damaged when handled in the classroom.

To further reduce the likelihood of breakage, certain steps can be taken during skin preparation to strengthen the vulnerable parts of the skin. The center piece of our efforts to "student-proof" our skins is to include a stout support stick in every skin. All vulnerable parts are then tied to this stick during preparation.

We begin by skinning and, if necessary, defatting and cleaning the skin in the traditional manner. A stick of suitable

size, usually a commercial wooden dowel, is chosen and finely sharpened at one end. The stick should be long enough so that the blunt end will extend 5-20 mm beyond the tail (or legs in long-legged birds) of the finished skin. On skins larger than about quail size, we make artificial bodies out of wood excelsior wrapped with string. On smaller birds, we use cotton. We begin by making a row of close wraps around the stick with string over the length of the main part of the body. We use white glue to assure a permanent bond between stick and string. A hard rather small central core of excelsior is then wrapped and tied over the wet glue-filled string and firmly tied to the stick, with extra ties at both ends of the core. This core and glue will prevent later separation of stick and body. The major portion of the body is then firmly wrapped and tied and shaped over the central core, using excelsior and string until a body of proper size and shape is achieved. To finish the body, cotton is wrapped and tied around the forward end of the stock to simulate the neck, leaving the sharply pointed anterior end of the stick bare except for a single layer of tightly wrapped string, again glued to the stick.

When skin and body are ready to be joined, we fill the skull cavity with white or yellow Play Doh (red and blue color will seep through and discolor the skin), and drive the sharpened end of the stick firmly through the skull cavity into the nasal bones as far as possible. We then tie the skull to the neck wrappings

and the stick with heavy button and carpet thread (finer thread on small birds), using a tie from the front of the skull across the top, and one on each side of the jaws back quite far on the neck wrappings so the skull is solidly tied back on the stick. Additional support will be provided when the Play Doh dries around the string and cotton wrappings on the portions of the stick that is embedded inside the skull cavity. The eye sockets are then filled with tightly wrapped cotton balls, and the skin is pulled back over the body. Wing bones and scapulars are hobbled with suitable thread in the traditional manner.

The skin is closed with stitches using the heaviest thread suitable for the size of the skin in question. Care is taken to use an ample length of thread in this final closure. After the skin is completely closed, the same, continuous thread is used to tie the tail to the support stick by driving the needle dorsally through the base of one side of the tail and returning ventrally across the other side of the tail base. Several half stitches will anchor the tail to the stick firmly. The same, continuous thread is then extended back along the stick to a suitable place, the thread is tied tightly to the stick using several wraps and half hitches, and this thread is then used to tie the legs and feet firmly to the stick. The loose ends of this same continuous thread are then double knotted about 10-20 mm away from the legs, and this free end of the thread is now available to receive the specimen tag.

On any skin with an excelsior body, a long needle with button and carpet thread is used to cross-tie the wings to the body. After final preening and wrapping for drying, the skin is cataloged and labelled. The final step in "student-proofing" our skins is to write the specimen number on the stick near the feet with permanent ink so that the specimen can always be identified and can be relabelled in case the label is lost.

With small birds using cotton bodies, the procedures are the same except that the bare stick is tied into the skull without having a body prewrapped onto it. Once the stick is anchored in place in the skull using Play Doh and tied, we then pull a triangle of cotton anteriorly through the skin in the traditional manner. This cotton is tucked in to form the body and the skin is closed. The tail and legs and label are all attached to the stick and skin as described above. Small skins with cotton bodies are inserted into protective plastic tubes when placed out for classroom use as additional protection.

In all of these preparations, it is important that the support stick extend beyond the most posterior portion of the made up skin, usually the tail, by at least 10-15 mm on large birds, and 5 mm on small birds. This will protect the ends of the tail from damage by sliding into hard objects such as specimen trays, tube ends, desks, etc. On large, long-legged birds we often tie the legs to the stick in two or three places with separate ties. Similarly, on very large birds with long wings,

we make two or three cross-body ties through the body to firmly anchor the wings to the body.

Making Exhibit Mounts From Previously Prepared or Field Prepared Specimens

by Greg Septon
Milwaukee Public Museum

In days of old, great numbers of birds were mounted as curios and ornamental displays. This was especially true in Victorian times. The problem with the great majority of these early mounts was that they lacked anatomical accuracy and, oftentimes, were portrayed in overly exaggerated or ghastly poses. In spite of this, these mounted birds were accepted by the public and became popular items.

Today, many of these same species of birds are threatened with extinction and, as such, cannot be legally acquired. This fact greatly increases the historical and scientific value of previously-mounted specimens. For museums intent on creating a public awareness of endangered species, nothing quite carries the point across as well as an interpretive exhibit centered around an accurately-mounted specimen. Unable to acquire fresh specimens for these exhibits, many taxidermists are now being called upon to either remount old specimens, or to relax and mount scientific study skins and alcoholic specimens (or "alcoholics," as they are called).

With this fact in mind, I have prepared the following material regarding the mounting of old bird study skins and alcoholic specimens. The methods and formulas described are those I've developed over several years of trial and error. Although the following material was prepared for and addresses the mounting of bird study skins, please keep in mind that much of it is also applicable for the mounting of alcoholics.

Before mounting a bird study skin, a taxidermist must determine if the skin is of a suitable nature to justify the ensuing amount of work. The old saying, "you can't make a silk purse out of a sow's ear" holds especially true with the mounting of preserved bird skins. If the skin is of inferior quality and was not properly prepared, if it is grease burned or damaged, or was collected during a period of moult, it should not be mounted.

When initially preparing an alcoholic, it is imperative that the specimen be thoroughly cleaned of all blood and body fluids, as the alcohol seems to set these and makes their removal from plumage very difficult. When inspecting an alcoholic prior to mounting, look for signs of this and avoid working these stained skins. Specimens preserved in alcohol, especially for long periods, show plumage discoloration and general fading. Very little can be done to avoid this.

Once a good study skin is chosen, attention must first be focused, as necessary, on preliminary relaxation of the legs, feet, wings, and skull areas. These generally take much more time to relax.

I prepare my soaking solution as follows:

SOAKING SOLUTION FORMULA

1 gallon water
1 pound salt
3 tablespoons mild, white, liquid soap (Ivory)
1/2 gallon alcohol
enough formic acid to bring the solution
to pH 3.5 (about 20 cc/gal.)

Once the solution is prepared, I dip strands of cotton into it and wrap these around the legs and, if necessary, the wings. These areas are then wrapped in plastic to keep the solution from evaporating. After about 8-10 hours, I remove the plastic and unwrap the cotton to determine how soft and flexible the legs and wings have become. The time necessary to relax the legs and wings varies with the size and condition of the skin.

Once soft, these areas can be further relaxed by injecting some of the soaking solution into them. I use discarded disposable diabetic syringes, as they have micro-fine needles which lend themselves beautifully to this type of work. When the legs and toes are injected, they will swell as they fill with the solution. The wing joints will also become thoroughly flexible after injection.

At this point, the entire skin is immersed into a bath of the soaking solution and allowed to re-hydrate. If placed in a refrigerator, a soaking skin may be kept for an indefinite period, although it is preferable to proceed with the actual mounting as soon as possible after the specimen is completely relaxed. While the skin is in the soaking solution, I check it frequently to determine its degree of flexibility.

When the skin becomes softened, the stitches can be exposed and cut, opening the skin for a more complete re-hydration. After removal of the filler material, I go over the inside of the skin with a fine wire brush to loosen the feather quills. The tendons (now softened) can also be removed from the legs; and the feet skinned by making an incision on the underside of each toe, allowing removal of the tendons and tissue. For small birds, this is not practical. In these instances, the toes are left intact and later injected with a 1:1 formalin/glycerin mixture.

With the skin now relaxed and pliable, it is first rinsed in water, then placed in the following baths to clean, insect proof, and degrease it:

BATH STEPS

1.) Wash and rinse 2-3 times in mild, white, liquid soap (Ivory) solution
2.) Edolan U Solution, 20 minutes[1]
3.) Acetic Acid Solution, 10 minutes
4.) Methanol 100%, 5 minutes
5.) Mineral Spirits 100%, 5 minutes

These same bath steps are used with all bird skins--fresh, dried, or alcoholic. After removing the bird skin from the final bath, it can be wrapped in clean, absorbant cloth or paper towels to soak up most of the mineral spirits. Afterwards, I normally blow out some of the mineral spirits with compressed air prior to tumbling the skin in a mechanical tumbler filled with hardwood sawdust. The sawdust soaks up the remaining mineral spirits and dries the plumage.

The final procedure involves fluffing out the feathers with a hair dryer at a medium heat setting. To ensure that the finished mount will have a smooth overall appearance, it is very important to make sure all the down and under feathers are completely dry, as they form the buoyant surface upon which the body feathers lie.

The actual mounting may now proceed. There are several techniques employed in this, and most taxidermists tend to alter a standard technique into one of their own. I find that when working with study skins, my methods of mounting vary from one specimen to another. The size, species, age, and condition of the skin will dictate how the actual mounting is approached. When working with preserved skins, the following must be taken into consideration.

1.) ELASTICITY--Most study skins do not have the elasticity or stretch of a fresh skin. This can limit the amount and degree of positioning the skin can withstand. Care must always be taken when working with an old skin, and a good sense of judgement as to how much "stress" a skin can take is essential.
Alcoholic specimens are often fixed in buffered formalin prior to immersion in alcohol. In these cases, the skin will have a rubber-like feel to it, and will be difficult to position without considerable pinning. I have found, especially with facial areas, that a complete thinning of the skin helps make positioning easier.

2.) OVER-MANIPULATION--With study skins especially, it is important to have a predetermined pose set for your mount and have the artificial body prepared to suit that pose, as over-manipulation of the skin can cause a loss of feathers or damage

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1Edolan U has recently been taken off the market as a hazardous material. A suitable replacement has not been found, as yet.

to the specimen. When the skin is mounted over the artificial body, a minimum of positioning should by needed.

3.) DRYING--Because of the fragility of some very old skins, the actual mounting process may be very time consuming, and premature drying of the skin may pose a problem. This is easily resolved by brushing the inside of the skin with a soft brush dipped in water that is mixed with a few drops of mild, white, liquid soap. The soap acts as a wetting agent, allowing the water to be thoroughly absorbed into the skin, keeping it supple.

4.) WIRING--When wiring the legs, wings and neck of a study skin mount, one must always keep in mind the stress limits of the skin. I've found that light gauge, flexible, soft-steel wires work best in all cases. Using the stiffer galvanized wire leads to problems in positioning and setting up the mount. With fragile skins, especially, the excess force needed to bend a stiff wire to position the legs will oftentimes damage the legs themselves. The key is to not incorporate anything into a mount that will cause unnecessary stress or make positioning a problem.

5.) PLUMAGE ADJUSTMENT--In cases where a specimen cannot be completely preened in one sitting, I keep them placed in a damp box or humidity chamber. The floor of the damp box is covered with silica sand and soaked with water and 5% phenol to prohibit the growth of bacteria. The damp box keeps the bird skin soft and allows me to work on the specimen at my leisure. When the plumage is all in place, the specimen is removed and allowed to dry.

Another problem that occurs when mounting old bird skins is the tendency for feathers to move about during drying. To alleviate this in some cases, I apply water-soluble linoleum paste to the inside of the skin and press the paste between the skin and the artificial body. This glues the skin to the body, locking the feather quills into position as they dry.

Sometimes, however, even with the use of the paste, there may be areas where the skin refuses to lay properly. In these cases, a good deal of pinning, carding, and wrapping with string is in order. Always use stainless steel insect pins, as both untreated and plated pins can rust and create a problem by leaving iron stains on the feathers and skin. While the specimen is drying, these pins are removed and checked daily to ensure that the feathers dry in proper position.

6.) FEET AND FINISHING--The preparation of bird feet often poses another problem. Bird feet are fleshy in life, and should be restored likewise. Filling the toes, which have been skinned open, with a maché compound and carefully sewing them back together allows them to be shaped from the outside as the specimen dries. All too often, a good bird mount is ruined by lack of attention to proper restoration of the feet and other fleshy areas, such as ceres, wattles, beaks, bills, and combs. Attention must also be paid to accurate setting of the glass eyes.

These six points should prove helpful when creating mounts from study skins or alcoholics. If approached with these suggestions in mind, and with patience, the challenge of mounting old study skins and alcoholics can be made a simpler and more enjoyable task.

[Compilers' note: For photographs illustrating several of the steps described above see: Septon, G. 1982. The Reunion. Lore, 34(4):2-11]

FREEZE DRYING VERTEBRATE SPECIMENS

Ronald E. Cole
Museum of Wildlife & Fisheries Biology
University of California, Davis

INTRODUCTION

Methods for preparing vertebrate specimens for museum retention have changed little in the past several hundred years. These methods include the classical study skin specimen (round or flat), a partial or complete bony skeleton (often accompanied by the skin of the specimen), and the fluid preserved specimen. Supplementing these preservation styles are special collections, including tissue sections, organs, eggs, karyograms, descriptive notes, etc. Because these three basic catagories of preservation (skin, skeleton, and fluid) have generally been adequate to answer many or most of the questions of past systematists (especially when combined with supplemental collections), scant attention has been given to developing new methods of specimen preservation.

In 1957, exhibit specialists at the Smithsonian Institution began experimenting with freeze drying as a method for preparing vertebrates for exhibit. This was a dramatic departure from the methods previously used in exhibit taxidermy, and as such it was (and is) viewed with skepticism by many, especially by classical taxidermists. In 1977, this author began adapting the Smithsonian's exhibit-based preparation methods for study skin archival. This paper discusses freeze drying as a museum tool (not as a method for preparing exhibits), with specific reference to observed advantages and disadvantages for the preparator. For a detailed discussion of preparation steps, technical information regarding freeze drying, and also discussions on other uses for freeze driers in museums (other than whole specimen preparation), please refer to Harris (1964), Hower (1967, 1970, 1979), Meryman (1960), and Romero-Sierra et. al. (1983).

Freeze drying (FD) is a technique by which an object is first frozen, then dehydrated by sublimation. Freeze drying has been instrumental in developing new ideas and products in foods and pharmaceuticals, but it has received little attention by the museum community. Because an entire specimen can be sublimated, the potential for multiple use, including retrospective analysis of parts, makes this technique attractive to researchers. Concomitantly, because the entire specimen can be sublimated, the possibility for specimen degredation from fat leakage and insect invasion makes this technique potentially hazardous for use in archival museums. Indeed, reports of insect damage to exhibit materials and to freeze dried sport hunting trophies have added to the clouds of suspicion surrounding this technique.

The present state of knowledge about freeze drying as a tool for systematic collections is not adequate to either endorse or condemn the FD process. This is due, in part, to the relatively young age of the process. Thirty years of casual use and some abuse by only a few technicians is not enough time to critically analyze the process or its potentials. As with any technique used in museums, critical analysis of actions and effects, through experimental design and documentation, is mandatory. This analysis of efficacy must apply not only to new techniques, such as freeze drying, but also to techniques currently in use, but untested through experimental design. The museum community, in reality, knows little about the efficacy of its archival methods. We have generally failed to record steps taken, materials used, and experimental design followed. Preservation methods have usually been chosen on the basis of tradition rather than efficacy, and the success or failure of preservation methods has not been well documented. The Museum of Wildlife and Fisheries Biology is a teaching resource museum of the University of California at Davis, and, as such, it is appropriate that we engage in basic research. We are also given the responsibility for developing vertebrate teaching collections. We view freeze drying as both a basic research question for museum scientists, and also as an efficient method of preparing specimens for classroom use.

THE PROCESS

A freeze dry apparatus consists of a refrigerated specimen chamber, a low-temperature water-vapor condenser, a vacuum pump, and an assortment of gauges, switches, and lights. While size of the machine is generally dictated by the size of an equipment budget, most freeze driers used in museums have an effective chamber size of 18" to 36" wide by 36" to 72" long. Within this size range, a commercially manufactured freeze drier costs between $10,000 and $18,000. Properly maintained, a freeze drier should give the user 10-15 years of service between overhauls (usually compressor or vacuum pump replacement), which reduces the purchase price to an affordable annual cost.

The standard freeze dry preparation style at the Museum of Wildlife & Fisheries Biology (MWFB) is to retain all or as much as possible of the specimen. Unless required for other research, all body parts including eyes and viscera are retained. A FD preparation begins as does a classic study skin preparation. That is, the specimen is first measured, then spot cleaned with distilled water to remove any blood or other stains. No cutting is required except for a very small lateral incision to facilitate sex determination. At this point, the preparator jumps ahead to what would normally be the last steps in study specimen preparation - positioning in the desired end-point pose, attaching the label, and lightly wrapping with either cotton wool or cheese cloth. The specimen is then completely frozen (but not flash frozen in liquid nitrogen, as this causes cracks due to

rapid temperature change and swelling), weighed, and introduced into the freeze drier. Specimens are periodically weighed (once each week at the MWFB), and when weight loss ceases, the specimen is removed from the freeze drier, accessioned, and entered into the archival collection which is housed in metal museum cabinets. While the FD process might take several weeks to several months (depending on the size of the specimen), the actual hands-on time in the freeze drier is reduced to a few seconds of weighing time each week.

Because FD is both a preparation method and a basic research topic at the MWFB, we record the standard morphometric information normally associated with our specimens (lengths of body, tail, wings, culmen, feet, plus weight, sex, age, descriptive notes, etc.), and we also record a historical sketch of the specimen from the moment it is received. This information includes date of death, date and condition upon arrival, length of time frozen prior to FD, storage temperature prior to FD, dates required FD, temperature and vapor pressure of FD apparatus time required for drying, and pertinent notes concerning archival, including fumigants (if any) used. While somewhat time consuming to record, we believe that this information is required to assess the potential of FD in the archival museum. In fact, background information on archival methods should be standard practice for all collections, both wet and dry. To this length, we are developing recording files for round specimens, skeletons, and fluid preserved material in addition to FD material. Data sets, forms, and information recorded will be discussed at a subsequent meeting.

The MWFB is a mid-sized teaching collection of approximately 3500 birds, 2500 mammals, and 5000 lots of fishes. Because of our relatively small collection size, each bird and mammal specimen is closely examined every month for signs of insect damage. In past years, the collections were routinely fumigated plus examined. However, we have recently ceased all routine fumigation in our collections and we now use the FD as an insect control device. That is, if any sign of insect presence is detected in a storage cabinet, the contents of the entire cabinet are freeze dried. We also FD all loan and classroom materials prior to their reintroduction back into storage. Following these procedures, we have never had a serious infestation of insect pests in our collections.

ADVANTAGES OF FREEZE DRYING

- Hands-on preparation time is reduced as freeze dried specimens require no evisceration or removal of other internal body parts.

- Difficult preparations (e.g., specimens which are molting, decomposing, formalin fixed, etc.) can be prepared with ease.

- All body parts are retained for retrospective analysis.

- FD specimens are much stronger (structurally) than conventional specimens because both the bony skeleton and the muscles are retained.

- It is relatively easy to maintain realistic form, feather arrangement, etc. with freeze dried specimens, as little if any disruption of parts is required.

- Shrinkage of soft parts is less than observed with air dried tissue.

- Unskilled preparators can easily and quickly be taught freeze dried preparation techniques.

- Because the freeze drying process takes place automatically, in a device which operates 24 hours each day, every day of the week or year, specimens can be prepared while the preparator is engaged in other duties.

Disadvantages of freeze drying

- The initial cost of a freeze dry apparatus ($12 - $18,000) is often beyond the means of many museums.

- Because specimens are often freeze dried without removing viscera and fat, eventual fat leakage is a major concern. (Note: at the MWFB, we have minimized this potential problem by choosing candidate specimens that are not considered high in body fats. However, this is merely avoiding a problem which should and will be the subject of a research project by our preparation staff).

- Because of the increased tissue mass of FD specimens (when compared to fiber-filled), there is a serious potential for increased interest by carrion-eating insects. (Note: This concern is based on the assumption that increased tissue mass increases the level of attractiveness to carrion-eating insects. This assumption should be researched).

- Because FD specimens normally contain all of their body parts, detection of insect pests living inside the specimen can be difficult for weeks or months after penetration. (Note: This is one of the most potentially serious drawbacks to FD specimens. Anyone considering FD as a tool for museum specimen preparations must be aware of this concern, and is invited to join with our staff in a collaborative research project to answer this question).

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THE CURATION OF AVIAN OSTEOLOGICAL COLLECTIONS

Diana G. Matthiesen, Collection Manager
Pierce Brodkorb Collections
Dept. of Zoology, Univ. of Florida, Gainesville, FL 32611

CONTENTS

INTRODUCTION

Zusi et al. (1982) in their analysis of the World Inventories of Avian Skeletal/Spirit Specimens (Wood et al. 1982), revealed the startling fact that roughly one-third of the bird species of the world were not represented by osteological specimens in any collection. In his review of the inventories, Woolfenden (1984) states that "these two volumes will have a greater impact on ornithology than any other book of the 1980s." In the same review, he comments that "The study of bird anatomy and paleontology has increased dramatically during the last few decades. I predict these fields of study will continue to grow."

The inventories, in addition to other factors, have greatly stimulated both the growth of avian anatomical collections and the research upon them. The ability to locate museum specimens using the inventories has made feasible research on fossil and subfossil birds that otherwise might not have been done because of the excessive effort required to locate the needed reference specimens. The SAPE (Society for Avian Paleontology and Evolution) has grown from a few dozen members at its First International Symposium in Lyon in 1985, to over 160 members, more than 100 of whom attended its Second International Symposium in Los Angeles in 1988. An even larger attendance is expected at its Third International Symposium in Frankfurt in 1992.

The growth of paleornithology has in part gone hand in hand with the growth of avian osteological collections, for skeletons are absolutely indispensable for this research. Curators are to be commended for their response in turning from preparing birds only as study skins, to the variety of preparations routinely done today.

THE "IDEAL" SPECIMEN

The phrase, "there's more than one way to skin a cat" had to have been coined by the first museum curator. Not only is there generally more than one way to achieve the desired end; but there is seldom unanimity as to the desired end. The bone of contention in skeletal preparation is the degree to which the specimen should remain articulated. There can be no one way to prepare a bird that satisfies all researchers, thus there can be no such thing as an "ideal" specimen.

Having examined skeletal material at many museums, it appears that the most common skeletal preparation is what I would call "semi-articulated": the specimen has been cleaned by dermestid beetles and is dismembered, but is otherwise still articulated. I will never cease to marvel at the way dermestids can clean a bird skeleton and leave it so beautifully articulated; but, whatever aesthetic value or novelty such a specimen may possess, those qualities are more than overshadowed by its near uselessness.

For paleontologists, zooarcheologists, and those engaged in the study of food habits (raptor pellets, carnivore scats, etc.), a completely disarticulated, tissue-free specimen is essential. The identification of bird bones requires that they be absolutely clean, because every detail (inter-muscular lines, foramina, rugosities, etc.) must be visible for comparison. Because birds, unlike other vertebrates, are so size-specific, measurements play a very important part in the identification process. Bones must be disarticulated if they are to be measured at all, and they must be tissue-free if the measurements are to be accurate.

We have all had the experience of non-ornithologists visiting our museums who express considerable disappointment at viewing drawers of study skins: they expected all the birds to be mounted in life-like positions. It's an able curator who can truly convince these visitors that, for research purposes, study skins (much less flat skins or shmoos) are more valuable than taxidermy mounts. I experience exactly the same situation with skeletons.

I'm not surprised when lay visitors are disappointed by the appearance of a boxed, disarticulated skeleton; for, to the non-specialist, it does appear to be a useless jumble. I am dismayed, however, when curators (whose own personal research does not involve the use of skeletons) adamantly insist that all the skeletons in their collections be semi-articulated. From the standpoint of research, an articulated skeleton is the equivalent of a taxidermy mount.

I've heard it argued that disarticulating a skeleton is a loss of information. I could equally argue that removing the skin from a bird is a loss of information. After all, a study skin (again, much less a flat skin or shmoo) doesn't very accurately represent the appearance of the whole bird; yet no one is insisting that all birds should be preserved pickled, perpetually frozen, or freeze-dried because skinning is a loss of information. We take for granted our familiarity with the shape of the living bird and the distortions inherent in a prepared skin.

Like skinning, the disarticulation of a bird skeleton involves an acceptable loss of information. Please believe me: all bird skeletons are put together the same way. The exceptions or ambiguities are so few that no more than a dozen or so well-chosen mounted or semi-articulated examples can serve as models for all other birds. Even the seemingly most obscure elements (e.g., ribs, toes, vertebrae, carpals) are individually identifiable. There is virtually no information irretrievably lost by disarticulation. Even if there were, the information lost in disarticulation is miniscule compared to the information unavailable in an articulated specimen.

There are exceptions to total disarticulation that I would cheerfully grant. For systematists and others, it is irritating to have the pterygoids, quadrates, and other bones of the skull disarticulated; so for them I would recommend keeping most skulls articulated. Bird skulls (crania) are extremely

rare as fossils in comparison to post-cranial elements, although loose quadrates are actually fairly common. At least some representative skulls in a collection do need to be disarticulated to allow thorough examination and measurement of quadrates and pterygoids. Also, the sclerotics and trachea do turn into a pretty useless jumble if disarticulated.

For skeletons, the most routinely useful specimen has disarticulated post-cranials and an articulated skull, sclerotics, and "windpipe" (hyoid + larynx + trachea + syrinx). Therefore, I prepare skeletons by first bugging the whole specimen with dermestid beetles and then using cold-water maceration for the post-cranials (and optionally the skull).

I have emphasized the argument for disarticulation of bird skeletons mainly because I know that most museums do not now prepare their skeletons in this way. I would urge that curators consider making the "semi-articulated" specimen the exception, while making specimens with macerated post-cranials the routine.

The methods I present below are couched in terms of preparing an entire skeleton without reference to the skin, but with the understanding that osseous preparation is the same for either a partial or complete skeleton.

The method I use has two goals: 1) to produce the kind of specimen described above and 2) to prepare it in the least possible amount of time. By "least amount of time," I do not mean shortest elapsed time from commencement to completion, but to the least amount of labor possible. For skeletal preparation, elapsed time and labor-intensiveness are very often inversely proportional.

SELECTION OF SPECIMENS TO SKELETONIZE

The inventories mentioned above have been of great help to curators in planning and managing the expansion of their collections both through additional collection and through trading. The usual object of acquisitive interest is a specimen that is adult, sexed, and fully documented; but, there are other, less obviously desirable, specimens that can be of considerable use.

It is the habit of most curators not to skeletonize juvenile birds because "they don't make good skeletons." To a point, immature bird bones can be identified, and immature fossil and subfossil specimens are quite common. Immature specimens are also common as prey in food-habit studies. There is a great need for osteologically-immature reference skeletons, most especially for older nestlings, fledglings, and subadults of known age. I have a personal need in my own research for first-year migrants taken in passage or on the wintering ground. (Avoid bugging immature skeletons: the dermestids will chew them to pieces; use maceration alone.)

Don't forego those busted-up road-kills. Broken elements can be very useful for determining bone thickness, cross-sections, and viewing internal features (especially of the skull). When road-kills are more broken than intact, consider saving the intact bones for your "grab bag." Over the years I have filled a good-sized box with the odd bones of mostly birds, but mammals, reptiles, amphibians, and even fish as well. It's a wonderful tool for teaching. Any odd parts, not normally worth saving or cataloguing, are good candidates for the grab bag.

Consider building an "element" collection: one where bones are stored by element, not by taxon. I can hardly overrate the value and usefulness of such a collection. A "bird-families-of-the-world" element collection is great for teaching, while for identification of bird remains, an element collection at the level of "genera-of-the-region" is needed. Once the remains are identified to the family/genus level, one reverts to using a standard "by-taxon" reference collection. Unsexed, dataless specimens are good candidates for an element collection, but it's very important that they be as complete and unbroken as possible. The element collection is definitely not the place for incomplete or broken skeletons: put them in the grab bag. Once you have established a good regional element collection, zooarcheologists will become heavy users of your collection.

Don't eschew those zoo birds or captive pet birds. They may be useless for ecological or systematic studies, but for identification purposes they can be just as useful as a wild bird. They often represent taxa that are very difficult to obtain in the wild.

Consider adding some "barnyard," "domestic," "restaurant," and "supermarket" skeletons to the collection as well (e.g., the carcass from your Thanksgiving turkey; quail ordered in a restaurant--I smuggle the carcasses out in my purse). Remains of these kinds of birds often occur as subfossils in historical sites and as the food remains of contemporary predators.

Even eroded, weathered, and gnawed-on specimens can be useful for the study of taphonomic processes, as examples of (what else?): erosion, weathering, and gnawing.

Taking into account all of the standard uses of bird skeletons plus the less common ones mentioned above, it's apparent that virtually any avian specimen has potential usefulness as a skeleton. Remember that putrefaction or mummification in no way reduces the quality of the finished skeleton, even though it may render the specimen unsexable. For systematic studies, knowing the sex of the specimen can be important; but for identification purposes, an unsexed specimen is often just as valuable. I would rather have a large sample of unsexed specimens (as one often gets with seabird wrecks, botulism outbreaks, tower kills) than a few sexed pairs.

Lastly, I know in the past that it has been the habit of some ornithologists not to skeletonize any specimen that could possibly be skinned, the value of a skeleton being considered so low as to be just one step above throwing the specimen away. Others would not even consider skeletonization of a skinnable specimen until there were at least five or ten pairs of skins in their collections. Please raise your priorities for skeletons, such that your second pair of a species is skeletonized. With the resurgence of interest in systematics and the burgeoning of research in the areas of paleontology and zooarcheology, there is a great need for these osteological materials.

DOCUMENTATION OF IDENTIFICATION

There isn't one of us who hasn't, at some time or another, misidentified a bird. If a bird is misidentified and then skinned, it is only a matter of time before someone corrects the error. If a bird has been misidentified and then prepared as a skeleton, it is entirely possible that the mistake may never be realized, the consequences of which are nightmarish. Bear in mind that, in general, the birds that are most likely to be misidentified whole are precisely the ones whose misidentification is least likely to be detected as skeletons.

Whenever possible, the specimen should be identified to the level of subspecies, because this determination is almost certain to be impossible based on the prepared skeleton alone.

Preparations that include a skin or spread wing with the complete or partial skeleton obviously provide adequate documentation of identification. For those preparations that do not include a skin, I strongly recommend some additional form of documentation. This documentation is especially important for those taxa where identification is known to be difficult (e.g., immature gulls, female ducks, fall warblers, etc.).

Some birds are so readily identifiable osteologically (e.g., Ospreys) that lack of documentation is not a problem, but osteological distinctiveness is the exception rather than the rule. If you know that a bird is osteologically unmistakable, then I won't argue with skipping the documentation, but otherwise please include it.

Probably the simplest form this documentation can take is to just make some note on the data tag to indicate the criterion or criteria used to make the species determination. This notation definitely need not be an entire description, just the character(s) that distinguishes that taxon from the one(s) with which it is most likely to have been confused.

Another method of documentation is to save the specimen's flight feathers (and possibly a patch of mantle or other diagnostic feathers). Used manila envelopes work well for storage, and soft-part colors and other information can be written on the outside of the envelope, including even a sketch of the

bill or feet. The reference feathers can be stored indefinitely in this form or eventually prepared further.

The flight feathers of one wing and half the tail can be glued directly into a manila file folder (a la the illustrations in Dwight's Gulls of the World) and the specimen data, written description, and sketches transferred as well. It's an enjoyable and educational project for students or volunteers. The file folder from one side can then serve as a guide to further identifications and as an instructional resource. A row of open folders showing all the stages of a gull species' plumage is quite dramatic--more so, I think, than a row of study skins whose remiges are barely visible. The flight feathers of the other side remain on file as documentation of the skeleton's identification.

The envelopes and manila folders can be stored inside standard office "archive" boxes inside museum cases. If the archive boxes are lined with a plastic garbage bag, the boxes can even be stored on open shelving. Arrow Star sells economical steel shelving meant to accommodate archive boxes. Called a "Transfer File Center," Stock No. LH2103 (72"w x 15"d x 60"h) holds 30 boxes (included) and costs only $93.75 (larger models hold up to 80 boxes). (Information on products and addresses of vendors mentioned throughout this paper are listed after the bibliography.) I have stored boxes this way for years and, provided the envelopes and folders are pest-free to begin with, I have had no problems with insects.

A surprisingly simple and effective method of documentation, unfortunately not usually available in the field, is to xerox the specimen! Even a monochrome xerox copy often shows enough pattern and gradation to serve for identification. It's only a matter of time before inexpensive table-top color copiers will be available. At UF, I sneak specimens, a can of Lysol Spray, and a Handi-Wipe into the department's xerox room in a tote bag. When the secretaries aren't looking, I xerox the specimens and then quickly clean the glass on the machine with the Lysol Spray. Given the kind of work I do, no one questions that I sometimes seem to reek of disinfectant.

The technology for desk-top-computer imaging of specimens is currently available, and it won't be long before it, too, will be economically feasible for museum use. The technology is also available now, and I'm sure I will see it used in my lifetime, for biologists to send specimen images from the field with their laptop computers, via satellite transmission, to the home museum.

Whatever the method, the important thing is to be sure that enough information is preserved so that the identification of the prepared skeleton is never in question, because once the specimen is roughed-out, there's no going back.

The value of a specimen is much increased by taking some additional data before the specimen is prepared: weight, wing chord, tail, culmen, etc. (see Baldwin et al. 1931).

ROUGHING-OUT: the "husked" specimen

"Roughing-out" simply means skinning the bird, gutting it, and cutting off the main muscle masses. The specimen is then dried for shipment, storage, and eventual placement in the dermestid colony. Dr. Jeanne Mortimer, a sea-turtle biologist, once saw a pile of roughed and dried carcasses in my lab and commented, "Oh, they've been husked." I can't imagine a more appropriate term for the preparation.

When training beginners to husk specimens, it's a very good idea to have an articulated or semi-articulated skeleton available for reference; so they can more readily see what not to cut through or pull off.

The specimen is first totally skinned. I begin by first pulling off the flight feathers and saving them for reference. Be careful not to pull off the distal phalanges of the wing with the primaries nor the pygostyle with the tail feathers. The tail feathers and primaries can almost always be reassembled in their correct order, though the secondaries usually cannot be.

The tail can be removed as two halves rather than as loose feathers. Pull off the upper and lower tail coverts so you can clearly see the bases of the rectrices and see, or at least feel, the pygostyle under the skin. Place a scalpel between the bases of the decks (the two central tail feathers) with the blade facing anteriorly. Carefully cut forward along each side of the pygostyle until each half of the tail is free from the bone. Most birds have a pygostyle that is flat, or at least flat-sided. Birds with long, strong, or elaborate tails tend to have more elaborate pygostyles: extremely so in woodpeckers, hornbills, toucans, some pheasants; somewhat so in passerines, cormorants, sulids, hawks, most galliforms. Feel your way gently with the blade. Once the feather bases are free from the pygostyle, pull laterally on each half of the tail and then cut away the tissue above and below the bases of the feathers, keeping the scalpel flat against them. Remove most of the adherent tissue and fat, leaving just enough to hold the feathers together in their natural arrangement.

The wing feathers can also be removed as a unit rather than as loose feathers, but it is a good deal more difficult and time-consuming. It takes some experience before you will get all, or even most, of the primaries in one piece. Begin at the bases of the secondaries on the underside of the wing, and from there skin the wing of all but the primaries and secondaries. Once all the skin and coverts are removed, begin at the elbow and slide the scalpel along the ulna freeing the bases of the secondaries from their quill knobs (the bumps on the ulna holding each secondary). Now work the scalpel flat against the bones of the manus freeing each primary as you go outwards towards the tip of the wing. It's helpful to have an articulated wing skeleton available for reference the first few times this is attempted.

The "articulated" tail and wing feathers need not necessarily be pinned and dried at this time. They can be folded and placed in a manila envelope just as you would loose feathers. The tissues will dry inside the envelope provided it is placed in a drying environment. Later, when the time comes for preparation of the flight feathers, they can be softened in water.

If dirty, feathers can be washed in cool water with liquid Woolite or in warm water with shampoo or dishwashing liquid. The feathers are then rinsed, rolled up and blotted with a terrycloth bath towel, and fluffed dry with a hair dryer. I personally don't like using powders of any kind (e.g., sawdust, cornmeal) to dry or dress feathers or skins. The feathers never really become totally clean again, as an examination under a scope will demonstrate. Also, the color of the bird seems, to me, to be subtly changed.

The skin can be cut off with scissors or scalpel, but often, after making some preliminary cuts (down the middle, around the neck), pulling the skin off in strips is easier and faster. Small birds, such as passerines, are most easily skinned under running tap water. Do a thorough job of skinning because at later stages of preparation you will regret every feather left on the bird. Dermestids will drag feathers into nooks and foramina leaving one to pick them out with needle and forceps. When pouring crocks, floating feathers tend to pull ribs and other light bones with them.

I highly recommend skinning the bird's feet. Not only will the specimen dry more quickly, but the dermestids will do a much better, faster job of cleaning the toes. Pedal phalanges, especially the basal and terminal rows, are highly identifiable; and, because of their density, are very common as fossils. Far too many skeletons have inadequately-cleaned feet, and failure to skin them is the main reason. The rhamphothecae and ungues (horny parts of the bill and claws) are part of the integument and can always be viewed in study skins. If left on the skeleton, they are obscuring important osteological features. If you do not skin the feet, then at least open them up with slits so that beetles and microbes have a better chance at working on them.

With a little practice, skinning the feet is not that difficult. Your most important tool is a pair of needle-nose pliers, plus either a scalpel (sharp) or pointed scissors (sharp). The real trick to avoiding frustration and pulled-off toes is in the careful preparation of the foot. Slit the tarsus along its entire length (both sides) and slit each toe, including the hallux, along its entire length (both sides). If the bird's foot is webbed, removing the webs accomplishes most of the lengthwise slits on the toes. Carefully isolate the skin on the hallux from the skin on the tarsometatarsus and make slits across the sole and top of the foot making sure that the skin on each toe becomes isolated. This sounds like a lot of trouble, but it's easy once it becomes routine. If all this slitting is carefully done, then each strip of skin pulls off the foot easily and rapidly using the needle-nose pliers. You may need to cut the skin free from the "ankle," but the rest should come

off easily. Pull only very gently on the skin of the hallux: on most birds, the first metatarsal is not at all firmly attached to the tarsometatarsus.

After skinning, open the abdominal cavity and sex the bird. If cornmeal has been used in skinning, wash it totally off the carcass and your hands before opening the abdominal cavity. Getting cornmeal inside the abdominal cavity can make it very difficult to determine the sex of a specimen. It's easier to get a good view of the gonads if the interior of the abdomen is completely exposed. With a small pair of scissors or sharp scalpel, cut around the abdominal area entirely: start along the caudal margin of the sternum, then up between the sternum and ribs, along the caudal margin of the ribs all the way up to the back bone then down the pubic bones, around the pubic bones and then along the caudal margin of the pelvis and the ventral surface of the tail, do the same on the other side, and lastly cut through the cloaca. Now the large flap of muscle and connective tissue covering the abdomen can be removed in one piece. Gently push the stomach and intestines towards the bird's right side and determine the sex, preferably taking measurements (length x width in mm) of the gonads (both sides).

Correct sexing of birds, especially reproductively-immature ones, requires first-hand instruction and some experience. The criteria are often described as males having two "beans" and as females having a single "bunch of grapes" on the left side. It's so easy to overlook an immature ovary and mistake the adrenals for testes that the beginner must beware of over-confidence in making sex determinations. I strongly recommend using a dissecting scope and bright light, especially for sexing passerines. Not only will the use of a scope reduce incorrect determinations, it's often possible to make a sex determination with a scope on a bird that appeared unsexable with the naked eye.

Don't automatically assume that because a bird is putrid that it cannot be sexed. It does take experience, but if a putrid bird is handled very carefully (not squeezed), and the abdomen is carefully opened (as described above), the outline of the gonads can often be seen, even in the putrid soup of the abdominal cavity. A scope and bright light are helpful, if not necessary.

Mummified birds can often be sexed, provided the abdominal cavity hasn't been maggoted. Soak the bird a day or two in water to rehydrate the gonads. I once received a large shipment of very roughly husked, unsexed seabirds (from a wreck of fulmars and shearwaters). About a third of the birds still had the dried kidneys and gonads present, so I was able to rehydrate and sex them.

Once sexed, the bird is then gutted. Gutting a bird can turn into a slow, picky, piece-at-time proposition (especially removing the lobes of the kidneys), or it can be done in one swell foop. Like rapid skinning of the feet, it's all in the preparation (plus a little practice).

With the tissue covering the abdomen removed, the next step is to free the trachea and the esophagus. Cut through and remove all the membranes around the furcula in the "hollow" of the neck. Separate the trachea and esophagus, freeing both of them along their lengths from head to trunk. Cut off the esophagus close to the trunk and to the head, taking care not to cut through the hyoid apparatus. Cut the hyoid apparatus free from the base of the skull and cut along the lower edge of the mandible all the way to its tip. Now reach down the trachea into the chest cavity with your fingers to feel for the syrinx. Holding the syrinx securely in your fingers, pull it free from the bronchi and lungs. Take care to put the pressure on the bronchi so that the syrinx is not damaged or pulled free from the trachea. With the "windpipe" (hyoid + larynx + trachea + syrinx) now separated, it can either be pickled or dried and bugged. There is little to recommend macerating the trachea--it really does turn into a useless jumble. (Believe it or not, I have found hyoid elements and syringes as subfossils.)

With your hand, reach up from the abdominal opening into the chest cavity along the underside of the sternum tearing the air sacs, vessels, etc. as you go. Work your hand forward and around the entire interior of the thorax freeing the organs from the bones by feel, taking care not to get stabbed by any broken ribs. There will be a tough mass of vessels and nerves to be freed in the axillae, and you will need to run a finger down between each pair of ribs freeing the lungs all the way to the back bone. If a thorough job is done of separating the organs from inside the rib cage, then it is usually possible to gut the bird entirely with one pull, including the lobes of the kidney. Grasp the freed organs firmly in one hand reaching all the way to the back bone with your fingers to get hold of the dorsal mesenteries. One firm pull should remove the lot. If you have not gotten a good enough hold on the supporting membranes at the base of the spine, you may leave the kidneys behind. With practice, this process is quite fast.

Puncture the center of the eye and express the fluid and lens from the opening. If the eye is large, cut away the entire iris, taking care not to cut the sclerotic ring. Not only does opening and emptying the eye aid in the drying of the head, it encourages the dermestids to clean the sclerotic rings, something they apparently do not relish. Don't remove the eyes from their sockets; they're too likely to get lost.

Just how much muscle is removed from the carcass depends on several factors, the most relevant probably being the conditions under which the specimens will be dried. Because drying is a problem in a humid climate, I generally remove as much muscle as possible. Other factors to consider are time available for husking, how hungry the dermestid colony is, and how fast you want the bugs to finish the specimen.

In larger birds, don't neglect removal of the muscles of the neck. It's tedious because care must be taken not to damage the intricate cervical vertebrae, but it's important to do if you want the specimen to dry rapidly.

At the very least, make slits in the fascia over the neck musculature to speed drying.

When skinning the knee, take care not to throw away the patella nor to cut through the thin cnemial crest of the tibiotarsus.

Remove the muscles at the nape of the neck until the gap between the atlas vertebra and foramen magnum is visible. Puncture the membrane over this gap to encourage drying and early access by beetles. If the specimen is very large and drying is going to be a big problem, the skull can be disjointed from the neck and the brains washed out with a stream of water and a probe or forceps, but this is not routinely necessary.

To speed drying, slit the skin between each pair of ribs, taking care not to cut through the uncinate processes. On larger birds, cut away the skin between the ribs entirely.

I do prefer to dismember the specimen, that is, I cut the wing free at the shoulder and the femur free at the hip. There's no trick to cutting off the wing, but to remove the femur, first cut away all the the surface musculature and tissue until the head of the femur is clearly visible in the acetabulum. To avoid an avulsion fracture of the head of the femur, don't pull outward on the leg. Instead, revolve the entire leg around the hip joint (in a parasagittal plane) several times. The ligament holding the femur in the socket will loosen, if not actually break, allowing the leg to be removed.

There are several advantages to dismembering the specimen. One of the most important is that with the shoulder joint opened, it can then be cleaned by the dermestids. For obvious reasons, the shoulder joint is the strongest joint in the body of most birds, and it is the structure most often left uncleaned in skeletal preparations. Having the shoulder joint bound with dried tissue is especially disadvantageous because the coracoid is otherwise the most identifiable of avian skeletal elements. The other main advantage is that a dismembered specimen will lie much flatter in the bugging tray, allowing shallow trays to be stacked on top of each other in the bug colony. The only disadvantage is that the wings and legs could get separated from the trunk during shipping and storage, so thorough wrapping with string is necessary.

If a toe or wing phalanx has gotten pulled off, stuff it into the eye, behind the eye in the eye socket, or through the foramen magnum into the cranial cavity to keep it from getting lost.

The husked specimen can now be bound together with soft string. Avoid binding too tightly; use more string if it seems insecure. A cone of 4-ply string would probably last the average museum a decade, and a twine-holder makes the job of wrapping much easier. Avoid thread, it can actually cut through a small bird's sternum. For a bird with sharp talons, place the feet inside the

rib-cage to avoid puncture wounds during subsequent handling. (Not an empty warning: I nearly lost a finger to blood-poisoning from the puncture of a hawk talon.)

It should go without saying that a durable, permanent, and waterproof label be attached to the specimen.

DATA LABELS

We've all experienced the frustration of seeing data lost because a specimen was improperly labeled. Study skins, professionally prepared in the field, usually have a finished, standard museum tag attached at the time of preparation and are thus the least likely specimens to have their data lost. In contrast, because most stages of skeletal preparation are messy, data tags are much more likely to be lost or destroyed. The other greatest source of data loss is perishable labels included in bags with frozen birds. Freezer labels often get wet and the paper and/or the ink dissolves into unreadability. For this reason, I catalog specimens when they come in the door, before they even go into the freezer.

Whether a specimen is given a final catalog number or merely an interim "working" number, a numbering system greatly reduces the loss of data. Upon receiving a specimen, the data are entered into the catalog and the "field" label is removed and put on file, cross-referenced to the catalog/working number. It may be very difficult to maintain the integrity of an entire data-label throughout all stages of skeletal preparation, but it is comparatively easy to keep a simple plastic number-tag intact and readable. It's also much easier to duplicate a simple number-tag when parts of the specimen are receiving different forms of preparation.

To make these number-tags a bit more informative, an alpha code is added. The alpha code system used is the one devised for North American bird-banders by Klimkiewicz & Robbins (1978). Obscure at first, these codes soon become quite familiar. For non-North American species, their system for creating new codes is used.

I use two kinds of tags, each including the catalog number, alpha code, and sex (if known), on a specimen that is to be skeletonized. One label is cut from a large sheet of matte mylar drafting film (available in any art supply store). This mylar label is totally water-resistant and can thus be used as the label in the freezer bag, in the dermestid tray, and right inside the macerating jar.

After the bird is husked, but before it is wrapped with string, the mylar label is placed inside the body cavity. Pressed against the underside of the sternum, the mylar label will usually stick and dry there. After binding the specimen with string, a paper string tag is attached to the specimen's leg.

The paper tag can be further protected by encasing it in a small (2x3-inch) zip-lock bag with the string trailing out the opening. If a putrid specimen is to be maggoted, both labels must be protected in such a manner. Maggots don't eat mylar, but something in their body chemistry or movements will rub the writing off the mylar label.

Use either pencil or india ink on the labels, never ball-points or felt-tip pens. A hard pencil (2-1/2 or 3) has the advantage that, even if the carbon is rubbed off, it embosses the mylar and the number remains readable. "Permanent" felt markers (e.g., "Sharpie") work well until they come into contact with grease or oil. Enough oil can wick up the string of a tag to dissolve "permanent" felt-tip markings into unreadability.

Never use metal wire in any way or in any stage of preparation. Most metals corrode or rust in a moist environment and will permanently stain bones. Corroding metal tags can inhibit dermestids and bacterial maceration. For these reasons and because they can corrode into illegibility, metal tags are not recommended, especially in marine environments (unless, of course, you can afford special, non-corrosive metal tags).

DRYING THE HUSKED SPECIMEN

The next step is for the husked specimen to be dried. In general, it is best to avoid subjecting skeletons to heat, especially dry heat, at any stage of their preparation. Dry heat, especially when applied too quickly, will split long bones and can cause the sternum to warp. Setting specimens in direct sunlight can cause splitting and warping as well.

The best agent for drying specimens is a high volume of cool, dry forced air. In the museum, this can be achieved by placing the specimens to be dried on racks in a fume hood, turning it on, and leaving the hood window open about an inch or two producing a strong draft of room-temperature air. An electric fan blowing across the specimens will also work, although the room will become filled with the odor of the specimens. Most specimens will be dry overnight.

In the field, these conditions can be very difficult to reproduce. Some form of screened-in box or cabinet is always useful. In an arid climate, there is little difficulty beyond keeping the specimens safe from scavengers (dogs, cats, coons, crows, ants) while they dry in the open air and shade. In a humid climate, there is little choice but to resort to some source of heat, but concentrate on increasing air flow while keeping the temperature as low as possible (preferably <140'F).

When on the road, specimens bound in string can be tied under the hood of your vehicle, and the hot air from the radiator fan will dry the specimen as you drive. Take care that none of the specimens actually rests against the engine

block or any other hot metal. In a humid climate, the specimens will rehydrate whenever the car is parked.

A gas or electric oven that can be set at a low temperature (120-140'F) will work provided air can circulate (set the door ajar). A home gas oven, with just the pilot light on, works very well and will usually dry specimens overnight. The problem is finding someone who will let you use their oven. When traveling, it's poor etiquette to press your host on this issue; and it's a rare friend, indeed, who will offer you its use.

On a cross-country trip, I once tried drying a husked, road-kill porcupine by hanging it in a nylon-mesh bag from the rafters of my hostess' garage. She never said a word, even though maggots rained down on the hood of her car all week.

Unless they are absolutely dry, husked specimens should not be placed in plastic bags because they'll get moldy. I wrap each dried husked carcass in a sheet of newspaper, secured with tape, and with an identifying stick-on label (number and alpha code) on the outside. Wrapping in paper reduces tangling and breakage, prevents loose parts from getting lost, and is added protection during shipping. Specimens can be stored in this form for years. It is also the form in which I most prefer to exchange specimens because it does not lock the other curator into any one form of final preparation.

THE DERMESTID COLONY

Dermestid beetles thrive in an environment that is warm, humid, and dark. Here in north Florida, it's possible to maintain an active colony outdoors most of the year with no more shelter than household garbage cans. For a few weeks in mid-winter, the colony must be sheltered on my porch because one hard freeze will knock a colony out. In California (where for five years I worked in the Museum of Birds and Mammals at San Jose State University) an outdoor colony survived year-round, but in general did less well because of the aridity and cool nights.

Indoors or out, I use 30-gallon garbage cans to house most of the colony. If outdoors, the area where the cans are kept must be fenced and secure from scavengers. The dried specimens are placed in trays or pans, which are then stacked at angles on top of each other inside the can.

I have found that plastic Rubbermaid drawer organizers (available in four sizes at supermarkets) make excellent dermestid trays. Their bottoms are flat and rigid, and they can thus be stacked quite high without tipping. For larger specimens (e.g., cormorant, peacock), I use cheap plastic dishpans. Most dishpans stack up five per can. They cost as little as $1 each on sale at discount stores. Look for ones with flat bottoms and try stacking them in the store before buying them. For tall birds (e.g., Great Blue Herons), the long,

narrow plastic plant-containers sold in garden stores are a very useful size. For the largest specimens (e.g., swans, pelicans) kitty-litter trays do nicely, though they will not fit in a garbage can. All of these plastic trays are easily washed for reuse, and they nest compactly for storage.

To bug something as large as an ostrich, use a plastic garbage can (dermestids don't like metal) by itself for the body and legs. Put the head and wings in trays to avoid lost bones.

I've tried using metal trays (the aluminum kind that frozen foods come in), but the dermestids deserted the specimens in them, especially the trays that had begun to corrode (from ammonia?). Dermestids seem to like cardboard trays best, but such trays don't last long because they are eaten up, and they can't be washed. Plastic trays are a practical and acceptable compromise. Dermestids sometimes chew holes in plastic trays, but these can be repaired with silicone cement or tape (e.g., duct tape). Avoid trays with seams or crevices, small bones can get stuck and lost.

To shelter the larger trays outdoors, I have found that a big airline dog-carrier (plastic with barred windows and door) makes a convenient shelter. The kennel is turned upside-down so that the windows slant downwards, not into the rain. Plastic garbage bags are taped to the top (formerly the bottom) of the kennel draping over the windows and door to keep it dark inside and to further keep out rain. Eighth-inch hardware-cloth had to be added to the open bars of the kennel's openings to keep out mice, lizards, and wrens.

Indoors, the larger trays can be arranged on steel shelving. Were I setting up an indoor colony and had some money to spend, Arrow Star sells a variety of industrial furnishings meant for "parts storage" (steel shelving with plastic bins) that would be excellent for use in organizing a dermestid colony.

Remove the string binding the specimen before placing it in the tray and remove the string from any tags or labels (easier now than later). Arrange the specimen in a tray and put a layer of fine-mesh cheesecloth over it. Use Grade 50 or 60 (24+ threads/inch) cheesecloth or several layers of a looser grade. Grade 10 or 20 is usually stocked by fabric stores, but finer grades will probably have to be special ordered (either through the fabric store or a supply house, such as Fisher Scientific). Cut the cheesecloth somewhat larger than the trays so that it trails over the edges and allows larvae to move from tray to tray. A long branch or piece of doweling extending from the bottom of the can to the top will allow larvae that have fallen to the bottom of the can to crawl back up onto the specimens.

A layer of cotton is placed over the layer of cheesecloth. The main purpose of the cheesecloth is to prevent cotton from contacting the specimen. The dermestids will pupate in the cotton and seem less likely to desert the specimen or the colony in search of a suitable substrate for pupation.

Dermestids don't normally chew into larger specimens unless they are exceptionally hungry or have no place to pupate.

Non-sterile cotton can be purchased in one-pound rolls for a fraction of the price of sterile cotton; also, be certain you get real cotton and not a synthetic substitute. Unroll a length of cotton and cut it into pieces the size of the trays using a large pair of scissors. The cotton is too thick as it comes off the roll, so peel it into three or four thinner layers. The cheesecloth and cotton can be reused, making it easy to introduce beetles to a new specimen. Under humid conditions, the cotton cannot be reused more than two or three times, apparently because frass in the cotton is prone to the production of ammonia and/or the cotton retains moisture.

In all other contexts, I cannot emphasize too strongly to never get cotton anywhere near skeletons. The fibers cling to the specimens, especially tiny ones, and are an incredible nuisance. Cotton fibers do not macerate away, and tufts of cotton can, like feathers, pull small bones with them when crocks are poured. Tissue paper is the best wrapping material for skeletons. Toilet paper is also acceptable; and because it comes ready-made in little squares, is very handy for wrapping individual bones.

How often the colony should be checked and how long it takes for a specimen to be cleaned varies enormously depending on many factors including the size of the colony, how well it's been fed, the weather, etc. Specimens can be cleaned overnight or take months, so it's a matter of always keeping track of the pace of the colony. In the main, dermestids do a beautiful job of cleaning specimens without damaging them; however, small bird skeletons can be badly damaged, so they must be watched very carefully. Monitor the progress of the specimen and remove larvae as they get larger, leaving only tiny ones to work on it.

Incidentally, specimens are dried to allow shipping and storing, to prevent mold and maggots, and to reduce putrefaction and odor. Given a choice, dermestids will preferentially feed on a soft, fresh carcass over a putrid or dried one. It's just that under natural conditions, carcasses don't stay fresh and dermestids don't appear to be able to compete with maggots for soft tissue. If you're in a hurry for a specimen, give it to the bugs husked, but not dried. Don't put a lot of moist material in the colony at once, however, or you'll end up with moldy specimens and possibly begin generating ammonia. Dermestids don't like cleaning moldy specimens, and ammonia fumes can kill a colony very quickly. Mold and ammonia can be a problem at any time if too much moisture or humidity builds up in the colony. Adequate ventilation and drier specimens will reduce this problem.

Indoors or out, spiders can become a problem in a colony. My solution is to carefully go through every tray manually popping the spiders and egg sacks (wearing surgical gloves!) whenever I'm routinely adding or removing specimens from the colony.

Outdoors, ants can be a very big problem, hauling away small bones as well as dermestid larvae and even building nests in the trays. I have solved this problem with "Tanglefoot," a sticky goo used by orchardists to paint around the trunks of trees to prevent infestations of crawling insects. It's available at larger garden and farm supply stores. Just paint a band of it around the outside of each can--and train yourself not to brush up against the cans.

Don't get pesticides anywhere near your colony. The only exception I make is for roach control: "Combat" trays are stuck to the outside of each can and at other strategic locations, but never inside the cans with the dermies.

Upon removal from the dermestid colony, specimens are placed in zip-lock bags. The "windpipe" and the sclerotics are individually bagged (the skull, too, if it is not to be macerated.) Because I routinely use at least two labels on a husked specimen, the paper tag is available to place in the bag with the parts not intended for maceration and the mylar label is placed in the bag with the parts to be macerated. Small zip-locks (2x3, 3x4, 3x5) are available from Greiger's or Cole-Parmer; for larger sizes, supermarket zip-locks are cheaper.

The bagged specimen is then placed in a freezer for a few days to kill any remaining beetles. Bugs can also be killed by brief exposure to heat; and if there is much residual moisture on the specimen, this will also dry it out. Do not bag moist or damp specimens without drying them or they will go moldy. Dried, bugged specimens can be stored in plastic bags indefinitely, awaiting further preparation or exchange.

DERMESTIDICIDE

I have never had a problem with a resurgence of beetles after freezing. Heat (120-140'F) will also kill dermestids and in a matter of minutes, and that low a temperature is usually not even harmful to feathers or skins. I no longer use chemical pesticides of any kind: a combination of heat, freezing, and "exclosure" (zip-lock bags, plastic boxes) has proven to be adequate, in spite of being in humid, buggy Florida. I have even kept study skins in zip-lock bags on open shelves in my office for years without their being attacked. Although it may sometimes seem to be the case, dermestids do not arise by spontaneous generation. Though they are fully capable of chewing through a plastic bag, I have never known them to infest specimens in clean bags from the outside.

There are additional advantages to macerating, rather than just bugging, skeletons. A bugged skeleton remains attractive to insects, but I have found that macerated skeletons (unless greasy) do not. It is very much less expensive, and certainly far more convenient, to house a skeleton collection in boxes on open shelving rather than in expensive, bug-proof museum cases.

With macerated specimens, storage on open shelving is entirely feasible, and chemical pesticides should not be necessary. If a boxed skeleton happens to become infested, it is simply popped in the freezer for a few days or weeks.

DERMESTID ALLERGY

Over the period of about a decade, I went from being totally unbothered by dermestids to being almost violently allergic to them, an experience that I know I share with others. The allergy takes two forms: a hay-fever-like respiratory allergy and a poison-ivy-like contact dermatitis. One reason I maintain my colony out-of-doors is that the fresh air makes working with the colony much more tolerable. I once examined under a dissecting scope the tiny blisters on my hands that resulted from working in the colony: in each blister was embedded a single dermestid-larva hair. To work in the colony I must now wear surgical gloves and mask and be clothed head to toe. My allergy to dermestids has gone so far that I now find it unpleasant to even work in the same room where a bugged skeleton collection is stored, and even brief handling of bugged skeletons produces a mild dermatitis. I may be an extreme case, but then I have processed literally thousands of skeletons over a period of about 15 years, and my research involves working with skeletons many hours each day. Such allergic reactions may await anyone who experiences enough contact.

Reduction of allergens is thus another advantage of macerated specimens over those that are bugged-only. For those skeletal parts that are not macerated, a thorough rinse in water (or water plus a little ammonia) is helpful.

COLD-WATER MACERATION

Cold-water maceration is the process of putting a specimen in a jar or crock filled with plain water and allowing bacteria to digest away all soft tissues leaving a thoroughly cleaned skeleton.

If a specimen comes out of the dermestids greasy, it helps to drill the long bones before it is macerated. Small electric drills that can be operated with a foot pedal (e.g., Dremel), can be found in hobby and craft stores and some hardware stores. Drill one hole towards the end of each greasy bone, being absolutely certain not to destroy any of the bones' topographical features.

The water covering the specimen is changed periodically, a malodorous process usually referred to as "pouring crocks" and an event often unappreciated by fellow occupants of your facility. At UF, I was able to solve this problem only by either pouring in the middle of the night or by doing the maceration at home on my screen porch. Although I still consider the other occupants of the building pretty wimpy to make such a fuss, I had to accept the fact that the ill will generated by pouring crocks wasn't worth being able to do it at

my own convenience. Keep this in mind when setting up where your maceration is to be done and don't underestimate how real this grievance will be perceived. Deodorizing sprays, such as those used by mortuaries, can reduce the odor problem.

By far the best deodorizing spray I have ever found is a vanilla-scented one made by Zep called Deo-V (roman numeral five). It comes full strength and is then diluted 1 oz. to the gallon and used in a manual, aerosol-pump bottle. Unfortunately, the smallest amount of Deo-V that can be purchased is a special-order, seven-gallon "pail" costing $133 (it comes standard in 55-gallon drums). Diluted, Deo-V works out to about 3.7 cents per quart, compared to Lysol Spray at over $5 per quart. Deo-V is definitely economical in the long run or worth getting together with another institution to make a purchase.

Incidentally, while there's no question as to the aesthetic objection to maceration odors, the odor itself is not a health hazard, though you will be faced with this argument. Contamination of wounds or food with crock microbes is obviously inadvisable, but even eating or drinking the contents of a crock would at most probably give someone the trots--if anyone could possibly conceive of doing such a thing.

I once macerated the carcass of a Thanksgiving turkey in an open bucket on my screen porch. After rotting for about three weeks undisturbed, I came home one day to find that my dog had emptied the bucket. To eat the bones, he had first to drink the water. Huskies have no class.

Next to the odor, the most frequent objection to maceration is the production of "white-stuff," a waxy, greasy, difficult-to-remove encrustation on bones. Prevention is the aim.

The amount of white-stuff on a finished skeleton is directly proportional to the amount of tissue on the specimen when it is placed in water. The fattiness of the specimen is also a factor. A major reason why I bug specimens before macerating is to have them as tissue-free as possible at the start of maceration. Specimens can be husked and then macerated directly, without being bugged, but doing so encourages white-stuff and also results in the skull, sclerotics, and "windpipe" becoming disarticulated.

White-stuff can further be prevented by using distilled water instead of regular tap water. White-stuff is similar to the insoluble scum that results from using soap in hard water. If you have a deionized water tap in your lab, use it for maceration--and any other process involving water and skeletons or skins.

White-stuff sometimes results from too infrequent pouring. As a rule of thumb, crocks should be poured whenever the water becomes densely cloudy. This

cloudiness may develop in as little as a few days initially to as long as several months towards the end of the process.

When pouring crocks I decant all liquid off the remaining pile of bones unless there is a lot of non-osseous tissue in the crock. In the latter case, empty the contents of the crock, little by little, into a large dissecting pan in the sink. Dilute the water in the tray and pour off the tissue, feathers, etc. carefully over the edge of the pan until the crock is emptied. Pour the specimen into the tray and rinse with several changes of water and then return it to the crock to continue macerating I pour everything out into a pan and pick out the skeletal elements for return to the crock. The partially cleaned skeleton in the crock should be rinsed several times before the crock is refilled and closed. There is no need to worry about the maceration restarting; everything in the crock (including the walls of the crock) is pretty well coated with bacteria even after the several rinses.

If crocks are poured too frequently, bacterial maceration will cease, and it's difficult to get it started again. Sometimes, dropping a bit of meat into a crock can stimulate bacterial activity if it has fallen off. Also, seeding a crock that is doing poorly with liquid from a crock that is doing well can stimulate activity. If a crock is proving very stubborn, I sometimes add a second specimen--of an entirely different type, e.g., a mouse or whole fish--to cause a resurgence of maceration. If you know your bones, separating the two later isn't difficult. Often, including some of the frass from the dermie colony in the crock seems to stimulate maceration. Often, including some of the frass from the dermie colony in the crock seems to stimulate maceration.

Don't be concerned if skeletons remain in crocks for months, even a year or more. Bones are not normally damaged by plain water, though there are a few undesirable conditions that infrequently do arise in crocks.

If crocks are exposed to too much light, green algae can grow. The algae will inhibit bacterial maceration, and the green stain on the bones cannot be removed without damage to the specimen. The algae will grow only when the water in the crock is clear, so presumably the specimen is nearly done when the algae does appear; therefore, it's best just to stop maceration if you see any green on the bones or the inside of the jar.

Bacterial maceration proceeds best at warm temperatures (80-90' F), but excessive heat will produce "red tide." I don't know what the organism is, but it severely inhibits maceration and the red stains on the bones are permanent. You might as well stop maceration if it appears. It is also difficult to eradicate once it gets started, so sterilize crocks that have been infected before reusing.

Rarely, a black, thready growth will appear inside the crocks intertwining with the bones. It does not seem to actually damage the bones, but removing it is extremely time-consuming. It's like tough, black spider webs, each bone

having to be manually picked clean. Maceration should be halted if it appears.

In general, long periods of maceration in water do not harm bones, but there are a few crock ailments that can rapidly ruin a specimen, so that their appearance is cause for immediate action. If you haven't had time to pour crocks for awhile, it's at least a good idea to glance over the crocks to be certain nothing nasty is going on. One of the real baddies is "clear jelly."

If at any time you can see a clear jelly (usually about 1/8" thick) coating the bones in a crock, pour the crock immediately and stop macerating. I have no idea what causes this condition, but it softens the bones into rubber.

Don't be concerned if a furry or jelly-like layer appears on the surface of the water in a crock, a rather common occurrence. Remove the layer from the surface, feeling through it carefully to be certain no floating bones embedded in the layer and thrown out, and pour as usual. Brush any floating, gunky bones clean; and put them back in the crock knocking the air out of them so they will sink.

One serious crock malady is "pink cream." If at any time a pink, creamy layer develops on the bones or on the surface of the water where bones are protruding or floating, immediately brush the bones clean and stop maceration. This "pink cream" (a fungus?) will severely erode and soften bones.

It can be alarming to find that the water in a crock, along with the bones, has turned coal black, but this condition is rarely harmful. Check the sternum and ribs, and if they seem at all rubbery, stop the maceration. The bones are usually clean anyway by the time the water turns black. If not, add fresh water and resume maceration. The specimen will turn white again when it is dried.

Some specimens received from zoos or pet stores will resist maceration because the animal was heavily dosed with antibiotics or some other drug. It may take 20 to 30 changes of water to leach out the drugs, but eventually the specimen will macerate.

Should you have had cause to interrupt maceration, a specimen can be finished by softening the tissues and manually scraping and brushing. The tissues can be softened by boiling, enzymes, enzyme detergent, and/or ultrasounding.

If, by listing all these maladies, I've made it sound like maceration is a troublesome procedure fraught with hazards, it is not. Most of the time, maceration proceeds smoothly; and a clean, ivory-colored skeleton is produced with a minimal amount of labor.

If an articulated skull is desired, but the dermestids have not eaten the rhamphotheca entirely away, macerate the skull just long enough to loosen it.

Pull firmly on the rhamphotheca each time you pour, so you can remove the skull from the crock as early as possible. It's usually ready on the first or second pour. After removing the rhamphotheca, toothbrush the rostrum clean. The ungues (horny claws) are usually loosened at about the same time as the rhamphotheca, so pull them off as well. Rinse the skull well (after optionally soaking in water with a little ammonia), and set it to dry with the pterygoids and quadrates still in place.

The most useful containers for maceration are tall, wide-mouthed, clear glass jars with plastic lids (e.g., Nescafe instant coffee jars are ideal). Metal lids rust and corrode and may stain bones and inhibit maceration. Colored glass jars will limit your ability to judge the progress of maceration or to spot the appearance of crock "diseases." Baby-food jars are a good size for passerines, but it may be necessary to put a piece of plastic wrap between the lid and jar because the lids tend to become rusty over time. The standard gallon glass "pickle" jar works well for specimens up to about the size of a large cormorant.

"Tupper-ware" type plastic containers can also be used: the size designed to hold a loaf of bread works very well for tall birds, such as Great Blue Herons. Don't stack large, water-filled, rectangular plastic containers on top of one another: the lids will break through at the corners.

For pelicans, a plastic 5-6 gallon garbage can works well. A 30-gallon can will be needed for a large ratite. Be sure to get a good quality plastic can, such as Rubbermaid, or it won't be able to take the strain of being tipped and poured without splitting open. Never use a metal garbage can or wash-tub, even galvanized ones. The galvanizing inhibits maceration and can leave a greyish-white residue on the bones. After the galvanizing corrodes off, the can will begin to rust, which doubly inhibits maceration and stains bones.

Before using any plastic container for the first time, fill it with several changes of water to leach out any chemicals remaining from the manufacturing process. Containers that have previously held toxic materials will inhibit or prevent maceration, so avoid used containers if you don't know what substance they've contained.

Never pour into an open sink. (I once had to open a sink trap to retrieve a bluebird skeleton that went down the drain when a jar slipped out of my hand.) I pour off crocks into an enamel dissecting pan. If any bones come out of the crock, they can be retrieved from the tray. I don't like sieves because they catch the gunk as well as the bones and because bones get stuck in the mesh and have to be picked out by hand, which is time-consuming.

On the last pour, the skeleton is poured into the drying tray, and it in turn is poured off into the sink tray. The point is to minimize any time-consuming transfer of individual bones with forceps. The metal or plastic trays that come with frozen foods make excellent drying trays.

The specimen is then left to air dry. As before, heat is to be avoided, but don't allow the drying process to take too long. If the specimen is not dried rapidly enough, it will mildew. The mildew first appears as tiny black specks, which cannot later be removed without damage to the specimen.

After the specimen is completely dry, it is stored in a zip-lock bag awaiting the next step in processing. It is possible to consider the specimen sufficiently cleaned at this point and to number and box the specimen at this stage, but I prefer to process specimens further.

ULTRASOUND

As they come directly from the crock, the bones of a skeleton are not entirely clean. The bones are at least covered with a slippery film of bacteria and may have white-stuff, dirt, or other deposits. Brushing each bone with a toothbrush is effective, but excessively time-consuming, and is usually damaging to smaller specimens. I prefer to process all skeletons through an ultrasonic cleaner, which renders them "squeaky" clean with a minimal amount of hand labor. Ultrasonic cleaning is extremely effective, and I have never known it to damage even the most delicate of skeletons.

I have a three-quart ultrasonic cleaner with heater and a timer (Cole-Parmer, Model #8851-34, $435). It's large enough to do a cormorant, but the long bones of taller birds (e.g., Great Blue Heron) have to be done one end at a time, so get the largest model you can afford.

For effective cleaning, some kind of surfactant (detergent) is required. I use Terg-A-Zyme, a laboratory-grade enzyme detergent made by Alconox. Avoid commercial enzyme detergents (e.g., Biz, Axion) because they contain bleach as well as enzymes. Use enough detergent to make the water feel soapy, about a teaspoon per quart of water.

Most specimens will be cleaned within 1/2 an hour. Two or three small specimens can be done at one time by putting them in individual jars or beakers (used plastic yogurt containers work well). The ultrasonic cleaner is more efficient when it is not overloaded. It is very important to rinse specimens in many changes of fresh water to leach them of all detergent. I do the first rinse in the ultrasonic cleaner and the rest on the sink counter.

Ultrasounding does not entirely dissolve white-stuff, but it does remove much of it, while loosening and softening the rest, making it easier to then brush off.

Ultrasounding with Terg-A-Zyme is an excellent way to clean owl pellets. The hair and feathers float to the top and the bones sink to the bottom.

BLEACHING

Don't. There is absolutely no need to bleach a reference skeleton. Stark white bones are difficult to visualize and to photograph. The bones are chalky, even crumbly and/or brittle.

Lay people apparently do find white bones more attractive, so for display, exhibit, or teaching, you may wish to bleach a skeleton slightly. If you do so, use an oxygen bleach (e.g., hydrogen peroxide, sodium perborate, "Snowy"). Never, ever, ever use a chlorine bleach (Clorox, Purex, sodium hypochlorite); it makes bones weak and chalky.

Hydrogen peroxide is available very inexpensively in liquid form at any drug store or supermarket. It deteriorates with age, so buy only a little at a time and store it in a refrigerator. It can be used full strength on the entire skeleton or painted on a stain with a brush.

Sodium perborate comes in powder form and is available from scientific supply houses. It is activated by heat and water. To bleach a skeleton, put it in a large metal beaker, saucepan, or pot and cover it with cool water. Heat the water slowly to a boil and then add the sodium perborate (about a teaspoon per quart). Allow the specimen to boil a few seconds and then turn off the heat. Let the specimen sit a few minutes, checking periodically to note the progress of the bleaching. The specimen appears darker wet than it will when it dries, so don't take the process too far. Pour off the liquid and replace with fresh hot water, avoiding any rapid temperature changes or the bones may split. Leach any residual chemical from the bones with many changes of fresh water.

Sodium perborate actually does more than just bleach the specimen, it has a cleansing and degreasing action as well, as does the boiling. Just bringing a specimen to a boil in plain water will bring a great deal of matter to the surface, including grease.

"Snowy" and other "color safe" oxygen bleaches available in supermarkets would probably work OK for bleaching skeletons, but I've never tried any of them.

As mentioned earlier, if maceration was halted before completion, the specimen may be finished by boiling (with or without detergent) and brushing. If one is, for some reason, in a great hurry for a specimen, it can be prepared entirely by boiling and brushing, in a matter of hours. Use of a household pressure cooker or autoclave will speed the process even more. It's a labor-intensive method that produces an inferior end product.

DEGREASING

I have found degreasing to be the most intractable problem in the preparation of bird skeletons. Under ideal conditions--and I wish I knew exactly what those conditions were so that they could be reproduced at will--the fat or grease in a skeleton will be digested away in the maceration process. Drilling a greasy skeleton before it is macerated, as mentioned earlier, can help microbes reach the interiors of bones to digest the marrow.

If a specimen has been macerated and ultrasounded with enzymes and is still greasy, organic solvents are probably the best next step. I have had actual experience with five different kinds of organic solvents: Stoddard, carbon tetrachloride, acetone, ethyl acetate, and white gas. The one that I have found most effective is Stoddard, a commercial dry-cleaning solvent made by Standard Oil. It stands to reason that all the qualities one would want in a solvent intended for cleaning woolen clothing (high solubility, high volatility, low residue) would be good for cleaning skins, feathers, and bones. Although carbon tetrachloride can be used as a dry-cleaning solvent, I have not found it to be nearly as good a degreaser as Stoddard. Acetone is an effective degreaser and is usually cheap and easy to obtain. (I do not presently know how to obtain Stoddard, so have been using acetone instead.) Ethyl acetate, like acetone, is an ingredient in nail polish remover and is also an excellent solvent and degreaser. All of the above organic solvents can penetrate skin (and respiratory membranes) and are thus very toxic. They should be used with solvent-proof gloves under a fume hood. White gas is not as toxic as the above, but it is a relatively poor degreaser in comparison.

As an alternative to organic solvents, there are several methods that work by mobilizing the grease with heat and carrying it away from the bones. Boiling in detergent, sodium perborate, or plain water will, like cooking soup, bring the fat to the top, so it can be poured or spooned off. Heating in an oven on paper towels will allow the grease to melt and run out of a drilled specimen. I've read that packing bones in dry plaster of paris and heating them will wick the grease out, but I've never tried it. Dry heat is more likely to split and warp bones than moist heat. Extreme heat and rapid changes in temperature should be avoided. The use of organic solvents is undoubtedly preferable to these methods.

YUCKY SPECIMENS

Not all specimens come to us fresh, especially if one is an ardent picker-upper of DOR's (dead-on-road) or DOB's (dead-on-beach). Instead of passing up those putrid or mummified specimens, remember that an unsexed skeleton is often just as useful as a sexed one.

Specimens found mummified have usually been maggoted and may or not have been thoroughly gone over by beetles. If there's much tissue on the specimen, try

having your beetles work it over. Give the specimen a stay in the freezer before placing it in the colony, or you may introduce some unwanted pests (e.g., spiders). Pull off the flight feathers and any large pieces of skin and body feathers.

If there's but a little tissue left on the mummy, it may be macerated directly. Remove all possible feathers before placing in a crock, because they tend to pull small bones with them when the crock is poured. It helps to dismember the specimen as well.

Depending on your aesthetic sensitivity, you may or may not wish to husk and dry a putrid specimen. At least try to skin and gut it, but if not, it is possible to just put the whole bird in a crock. The first pour will be a dilly. As above, make that first pour a gradual one, so the crock liquid becomes much diluted, and the feathers, skin, guts, and bones are easier to separate. (This is only slightly less unpleasant than husking it in the first place.)

Another way to deal with a noxious specimen is to let the maggots work it over first. Just put the specimen in a dishpan, cover it loosely, and let nature take its course. A cage-box may be necessary to exclude scavengers. Roofs can be excellent places for maggoting specimens, provided you can secure the specimen from rats, crows, cats, gulls, etc.

San Jose State University (a downtown, urban campus) once received a putrid Elephant Seal, and the mammal technician decided to put the specimen on the roof of the science building to let the maggots work it over, rather than husking it. About two weeks later, I got a call from Buildings and Grounds asking me where she had put the seal. I didn't know exactly, so the supervisor and I went up on the roof together to find it. The technician had placed the seal in the shade, which would have been a good idea, except that it was the shade of the intake funnel for the building's air conditioning system.

I once picked up a putrid, road-kill skunk and, not wanting to husk it, put it in a loosely-covered plastic dishpan in my backyard. First the maggots worked it over, then the dermestids (et al.). When the insects were done, I filled the dishpan with water to allow it to macerate. I waited for a rainy day to do the first pour, so that the liquid would be immediately washed deep down into the sandy soil. Unfortunately, the liquid did not percolate straight down, but followed the contour of the slope of my yard--from back to front. It took about three weeks of summer thunderstorms for the bolus of skunk water to get flushed under the house and out to the street, it's position at all times being discernible.

In the case of seabird wrecks or disease outbreaks, it's possible to receive far more specimens than one can process (or find room to freeze) at one time. Rather than throw specimens away, there is a way to save them. It's not a

desirable way to prepare skeletons; it's just better than trashing them. Triage the freshest specimens into the freezer or set them aside for immediate husking. For the rest, put each specimen (opened and sexed, if possible) in a zip-lock bag with a mylar label, fill the bag with water, load the specimens into a plastic garbage can, and fill the can with water. Obviously, the can will have to be stored outdoors. As time permits, each bird can be poured from its bag into an individual crock to finish maceration. Hardly an ideal method, but better than passing up what may turn out to have been a one-time-only opportunity to acquire a big sample of a species.

GLOVES AND MASKS

The average person has an inordinate fear of disease from dead animals, while we museum workers tend to be overly cavalier about the risks involved in preparing specimens. Compared to mammals, birds present a relatively low risk for transmission of diseases, but anyone taking those risks should not do so in ignorance. Anyone working with animals should read Irvin and Cooper's (1972) summary of health hazards and scan a copy of An Outline of the Zoonoses (Schnurrenberger and Hubbert 1981). Each of us has the right to take whatever risks we please, but, for reasons of liability if not decency, we should be certain that volunteers, students, and others who may work for us on a casual basis are protected with gloves and masks and are informed of the hazards.

A stay in the freezer can reduce the disease potential of a specimen by directly killing disease organisms and ectoparasites. This is more important for mammals than birds, but I'm always more comfortable preparing a specimen that has been long frozen than one that is freshly dead.

The most common risk is infection from a cut or puncture, which can be trivial to life-threatening. Your best protection against cuts, aside from being careful and not working with dull tools, is to wear surgical gloves. Good surgical gloves can take a surprisingly strong glance with a scalpel and not give way. I had to nearly loose a finger to blood poisoning before I became religious about wearing gloves. (Besides, the smell of carrion on your hands is hard on your love-life.)

If you have an infant child or an ill or elderly family member (i.e., anyone with a compromised immune system), or are sexually active, you should give some thought to the consequences of coming home with your hands and nails impregnated with possibly virulent microbes. If you've been pouring crocks or handling putrid carcasses with ungloved hands, you should definitely not be the one elected to prepare the egg-salad sandwiches.

It's a good idea, at the end of any prep session--gloved or not, to wash your hands thoroughly with a disinfecting soap and a scrubbing brush. The brushing is important; it makes a significant difference in how clean your hands become. Betadine is a very widely used surgical antiseptic that is effective

and non-irritating and is available in drug stores and some supermarkets. For hand-washing, it is most usable in the form mixed with soap: "Betadine Skin Cleanser," about $5 for a 4-oz. plastic squeeze-bottle. For disinfecting and dressing wounds, use "Betadine Solution," about $4 for an 8-oz. plastic squeeze-bottle.

Since my episode of blood poisoning some ten years ago, I have followed my physician's instructions regarding the treatment of cuts. I have not had a subsequent infection, in spite of some nasty cuts while working on some pretty gross specimens. His instructions were: Immediately pull off your gloves and make the cut bleed profusely. Wash your hands thoroughly with Betadine Skin Cleanser in running hot water, continuing to encourage the cut to bleed. Blot your hands dry with a clean towel, but don't touch and contaminate the cut itself. Hold your hand up to encourage the bleeding to stop. If necessary, apply direct pressure with a sterile dressing to stop the bleeding. Put some Betadine Solution on a sterile dressing and then bandage the wound well. Use a fresh, intact pair of gloves to resume working. Keep the wound clean and bandaged until it heals; if the wound becomes red and hot, seek medical attention immediately.

Most people hate the thought of wearing gloves, principally because their experience with them is limited to the cheap ones, the kind that come only in "small, medium, and large" sizes. Go to a surgical supply house and get real surgical gloves, the kind that come in half-sizes. If you don't know your glove size, you'll have to buy a few pairs around your size to find just the right one. The average woman wears a size 6 or 7, the average man about an 8 or 9; different brands vary somewhat. Too tight a glove will restrict blood circulation and become very uncomfortable, too loose a glove will be cumbersome. With a properly fitting glove, you will forget it is even on, and there will be no loss of feel or dexterity.

Good surgical gloves cost from $.75 to $1.50 a pair depending on the brand and quantity purchased. Brands vary considerably in their comfort and durability, so try several. My favorite is "Micro-Touch." You can get several wearings out of a single pair with a little maintenance: At the end of each wearing wash the gloves clean, blot them dry with a towel, and dust them with baby powder. Remove the gloves inverting the fingers as you go. If a finger remains folded, blow into the glove like a balloon until all fingers are unfolded. Blot the inside of the glove dry and dust with baby powder. Sunlight and heat deteriorate gloves, so store them in a cool, dark place.

Semi-rigid, formed masks are readily available in paint stores, but I much prefer real surgical masks: soft, flexible, pressed-fabric masks with a thin metal band in the upper edge to form a custom fit over the bridge of the nose. Not only are these masks more comfortable, but, if you wear glasses, they are less prone to steaming up. Bothersome at first, it isn't long before you don't even notice that you have it on.

When pouring crocks, one occasionally gets splashed in the face with the crock liquid. In spite of wearing glasses, I have twice contracted conjunctivitis from splashing crock liquid in my eye. If at any time you get crock liquid or other putrid matter in your eyes, wash them immediately with clean water and seek medical attention. I now keep conjunctivitis medication on hand and use it prophylactically (you will need a prescription from your physician), whenever something putrid gets in my eyes. I haven't had any problems since I began taking this precaution.

LABELING

It's important that every bone of a skeleton be labeled with its catalog number in permanent black ink. This practice is essentially universal, though I do wish museums would include their acronym with the catalog number (e.g., MVZ-19378, CM-2343, etc.). Doing so would greatly facilitate return of those odd, mislaid bones that one finds in collections.

The cleaner a specimen is, the more readily it will take a number. Macerated specimens are easier to number than bugged ones, and specimens that have been ultrasounded are easiest of all. If the bone still resists numbering, rubbing first with a moistened Q-tip, then with a dry one, will usually suffice to clean a spot for numbering.

Numbering skeletons has got to be one of the most mindless and tedious of museum tasks. I usually do it at home in the evenings, while watching, or rather listening to, television. The chore is often relegated to students or volunteers, who may not appreciate the importance of just where numbers are placed on each bone. It cannot be emphasized too strongly that numbers never be written over any discernible feature (line, foramen, rugosity, etc.) on a bone. It makes it surprisingly difficult, if not impossible, to see the features so covered. Using too large a pen size is often partly responsible for the obliteration of features.

My preferred Rapidograph pen size for most numbering is a 4/0, with a 3/0 for large birds (swans, etc.) and 6/0 for small birds (passerines, sandpipers, etc.) and for the smaller bones of larger birds.

Fine-point Rapidograph pens can be difficult to keep working, but the following should facilitate their use and extend their life: Keep the pens in Koh-I-Noor's "Dry Double-Seal" modules. Not only do these modules keep the points from drying out, each module acts as a pen holder, so the pen need not constantly be capped and uncapped during use. The modules interlock, so that a bank of various pen sizes forms a single unit.

At the beginning of each day's use, before shaking the pen to get it started, immerse the point briefly in an ultrasonic cleaner. Small, desk-top ultrasonic cleaners are available in art supply stores for about $30, an investment

that will soon pay for itself in replacement points. Post-mortems on 6/0 pen points has revealed the primary cause of mortality is shaking the pen when the wire is dried and stuck in the point. The wire is pulled free from its plunger and that's the end of the point.

When a pen becomes so clogged that ultrasounding of the tip alone will not get the ink flowing freely, it's time to dismantle the pen and ultrasound it completely. A little Ivory Liquid in the water will do the job; there's no need to waste money on special "pen-cleaning" soaps. Larger pen-points can be taken totally apart, removing even the internal plunger. Never dismantle a 6/0 or 4/0 pen to this extent; it is impossible to get the fine wire of the plunger back down into the pen-point.

Koh-I-Noor "Universal" (3080-F1) waterproof India ink works well with larger pen sizes, but tends to clog in smaller points. "Rapidraw" (3084-F1) is a densely-black, permanent, latex base ink that is less clogging than Universal ink. For 6/0 pens, I recommend "Ultradraw" (3085-F); it isn't as dense as Rapidraw, but is the least clogging of the three kinds of ink. Always buy inks in small quantities (3/4 oz.), because they deteriorate (separate, thicken) with age.

The next most common cause of pen-point demise is getting the tip clogged with matter from the surface being numbered. If bones (or fossils or subfossils) are first cleaned with an ultrasonic cleaner, this kind of clogging is virtually eliminated. Dropping a pen point-down on the floor will ruin it. Avoid doing this.

Fine points that manage to survive other sources of mortality will eventually wear down and become difficult to write with (blobbing, scratching, skipping). Check the tip under a scope to see if it has been damaged or worn. Jewel-tip pen-points are much more wear-resistant and are noticeably smoother and easier to write with than regular stainless-steel pen-points. However, the jewel-tipped pen-points are just as prone to clogging and to having the wire separate from the plunger, so they may not be worth the extra expense (about two to three times the price).

It's essential that bones in an element collection be individually labeled as to taxon. The first element collection I built was a small, regional one of birds and mammals for zooarcheological research at San Jose State University. I labeled each bone with the genus name, but soon came to find this impractical. Not only are some genus names too long to fit on small bones, the names are not sufficiently stable. I now use the four-letter alpha codes (as mentioned earlier), which are shorter and, because they are based on the common name, more stable. If known, the sex of the specimen is indicated as well.

I found it necessary to label paired elements with their side, partly for teaching purposes, but mostly because students will not put the elements back

in the correct boxes if they're not so labeled. Phalanges receive their digit and phalanx number as part of the side (e.g., R-III-2).

Labeling the manual and pedal phalanges with their side, digit, and phalanx requires that one have a model specimen for a guide. Whenever I prepare the first specimen of a taxon, I hold back one bugged foot and wing to serve as a model for labeling the macerated phalanges of the other side. Then the bugged wing and foot are macerated and labeled using the labeled loose phalanges as a guide. It isn't necessary to have a model of every species for labeling. Certainly within a genus, and usually within a family, all members are sufficiently alike for one to be a model for the others.

I have found that it is extremely useful to include the alpha code, sex, and side on all specimens, not just those intended for the element collection. Anyone who has ever worked on identifying bird bones has been in the position of arranging open skeleton boxes on a work table constantly looking back to check the box labels to see which number belongs to which taxon. Having the alpha code and sex on every bone makes working with reference skeletons very much easier. It also reduces instances where bones are put back in the wrong box. It's helpful to put the alpha code on the box label as well.

There is another reason why it's useful to put alpha codes, etc., on every specimen, and not just those specimens intended for the element collection. It turns out that the composition of an element collection, especially while it's being built, is actually very dynamic. If all skeletons are fully labeled, it's very easy to move them in or out of the element collection; one is not forced to make a decision at the outset as to whether a specimen is permanently relegated to the element collection or to the reference collection. You may, for example, have your only specimen of a key genus or family in the element collection. If that specimen is incomplete and you later get a complete one, you will want to switch the complete one into the element collection. Also, if your first specimen is sexed and the second turns out to be unsexed, you will want to switch the unsexed specimen into the element collection. As you work on bird remains from different parts of the world, it's worthwhile to adjust the composition of the element collection for the region under study.

Incidentally, people often ask me how to learn to identify bird bones. My best recommendation as to how to become expert in recognizing bird bones is to build an element collection, because complete sorting and labeling requires identification of every bone in a skeleton to element.

Numbers can get rubbed off bones, especially those specimens used in an element collection or for teaching, because they are handled a great deal. Clear nail polish can be used to paint over the number on a bone to help keep it from getting rubbed off. If you buy a cheap brand at a discount drug store, it's very inexpensive, and because it comes in little bottles with their own brushes, it's very convenient to use. If, for some reason, you wish

to remove the nail polish, acetone on a Q-tip will do so, as will soaking the entire specimen in acetone.

The pubic bones of most birds are thin and fragile and tend to get broken off the pelvis, so they should each be labeled individually. It's a good idea to glue the pubes to the ischia distally, where they are attached in life by ligaments. This is anatomically more correct and will help prevent the pubes being broken off. This is done at the time the skeleton is labeled. Use Duco Household Cement, available in any hardware store and most office supply stores. Not only is Duco clear and fast-drying, it is soluble in acetone and can thus easily be removed if needed. Avoid any glues that cannot be unglued. Some birds (e.g., raptors) have pubes with no osseous connection with the innominate; these are left loose and not glued on.

HOUSING AND ORGANIZING THE REFERENCE COLLECTION

Avian skeletal collections are universally stored in taxonomic order, one specimen per box, in museum cases. Both the boxes, usually custom made, and the museum cases have become extremely expensive. It is very, very much more convenient to use an osteological collection that is housed on open steel shelving, which costs a fraction of the price of museum cases. Given the budget-tightening we all seem to be facing, this is definitely a place to conserve funds.

I strongly recommend organizing your skeleton collection in the nomenclature and classification of the "inventory" (Wood et al. 1986), whatever your personal taxonomic preferences may be. The Brodkorb collections, both skins and skeletons, have been converted to this order. The "inventory" represents the best opportunity we've had to introducing some uniformity to the organization of collections. Making exchanges is easier, as is keeping up your own inventory. It's also makes it easier for visiting researchers to work in your collection if its organization is a familiar one.

The identification of bird bones can also be facilitated by building an element collection (discussed in the next section) and by organizing the minor elements in the boxes of reference skeletons.

In virtually every fossil or subfossil fauna that I have identified, at least a few taxa have been added to the species list on the basis of "minor" elements (e.g., carpals, phalanges, vertebrae, radii, etc.). While these minor elements may not be useful systematically (i.e., for the description of new species), identification of all taxa is important for paleoecological analysis, and identification of all skeletal elements is important for taphonomic analysis. Yet, these minor elements tend to be ignored because identification seems insurmountably difficult. This difficulty is in part due

to the fact that the small bones of a bird tend to end up as an obscure-looking jumble in the bottom of the box.

Bones of like kind can be bagged in small (2x3, 3x4, 3x5) zip-lock bags. I bag the following: sclerotics, hyoid/larynx, trachea/syrinx, vertebral ribs, sternal ribs, manual phalanges, pedal phalanges (except ungulars), ungular phalanges, caudal vertebrae, and a "miscellaneous" bag containing the carpals, patellae, pygostyle, atlas, axis, metatarsal I's, and any other small, loose bones (pubes, intertarsal sesamoids, quadrates, nuchales, etc.). Cervical and thoracic vertebrae are restrung.

Bones are identifiable to the degree to which they have features. Vertebrae have many features and are thus very identifiable; however, loose in the bottom of a box, it's a daunting task to work with them. Vertebrae can be put back in order and restrung. Each vertebra is unique and, with practice, getting them back in order takes just a few minutes. I also label each vertebra with its order number at this time.

Thread the vertebrae onto a piece of twine beginning anteriorly, and then tie off the loose end. To tie off the end, I use a clear plastic, tri-partite bead. These beads are small and unobtrusive, yet resist getting pulled back through the neural canals. These beads are available very cheaply (a few dollars per thousand) in craft or hobby supply stores. Also available at craft stores is braided twine in several weights and in a natural, ecru color that is very close to the color of bones. Braided twine doesn't unravel like twisted twines, and is less prone to tangling. Don't use thread; it's much too tangly. Push the vertebrae along the twine until they come together up against the terminal bead. Cut the twine about eight inches longer than the vertebral column, to give some slack so that individual vertebrae can be examined easily. A one-inch diameter, metal-rimmed key-tag (available in office supply stores) is tied to the anterior end of the string. The tag is labeled with the catalog number, alpha code and/or scientific name, and sex. Write the label so that it reads upright when the hole is downwards (you'll see why below).

Because the atlases, axes, and notaria (fused thoracics) are so unique, I keep them loose and do not string them with the other vertebrae. They are labeled with their alpha codes. A tiny white tag is placed on the strung vertebrae in the place of the missing notarium to indicate its absence from the string. With the vertebrae strung and small bones bagged, a boxed reference specimen is far easier to use and much easier to pack for shipment.

Specimens are also easier to use if they are not forced into boxes that are too small. I know boxes are expensive and space in a museum is at a premium, but it's difficult and time-consuming to work with specimens that are jammed into small boxes. Visiting researchers almost always have a very limited time to work in a collection, and it's very frustrating to spend 20-30% of that time struggling to extract the needed element and to repack them in boxes that

are crammed too full. The box should be large enough to allow one to find the element needed without dumping out the entire contents of the box. Tightly boxed skeletons are often damaged because someone lacked the patience to repack them properly. From the standpoint of ease of use, the best skeleton box is larger and lower than the boxes used in most collections.

If at any point the cost of boxes has caused a museum to stop processing skeletons, worse yet, to discard specimens, there is an interim measure one can take to at least insure that valuable specimens are not passed up.

Prepared skeletons can usefully be stored in zip-lock plastic bags. Just inside the lip of the bag, a stick-on label is placed, containing catalog number, taxon (scientific name and/or alpha code), and sex. These bags are then stored in clear plastic sweater boxes (11"w x 14"l x 7"d), available inexpensively ($3.49 each) at discount stores (Pic-N-Sav, Wal-Mart). The boxes are stored on open shelving. 5x8 index cards, listing the contents of each box, are placed inside and are visible through the ends of the boxes. Larger specimens are bagged and placed in office archive boxes (on the same kind of shelving as used for reference feathers).

Skeletons stored in zip-locks take up a fraction of the space of boxed skeletons. Getting a specimen in or out of a zip-lock bag is very easy; one is never faced with the frustrating task of trying to fit a skeleton into too small a box. Bagged specimens of the same species, genus, or family can be placed together in a larger zip-lock, making them easier to find inside the plastic box. Bugged specimens awaiting maceration can also be stored in zip-lock bags this way, thus preventing the soiling of the inside of an expensive, custom cardboard box.

I began storing specimens in zip-locks as a stop-gap measure until money was available for custom-made boxes, but frankly, it has turned out to be so useful and so economical, I'm not sure the added expense of individual boxes is worth it. I don't suppose I'll convert anyone to abandoning the storage of skeletons in individual boxes, but I do hope more museums will revert to storing skeletons on open steel shelving.

HOUSING AND ORGANIZING THE ELEMENT COLLECTION

A well-organized element collection, in conjunction with a well-organized reference collection, can can enormously speed the identification of avian remains.

In an element collection, most of the bones are kept loose in open trays placed in shallow drawers. One kind of element is in each tray. Paired elements are kept separated by side. I do not recommend keeping only one side of paired elements in the collection just to save trays and space. Many people, including myself, find it very difficult to make mental "mirror

images" to identify bones. The wasted time, and increased likelihood of error, are not worth the small savings in trays and drawers.

The best way to house an element collection is in cabinets with shallow drawers. For small specimens, cabinets like those used for instruments in dentists' offices are ideal, but expensive. Sometimes, one can find small cabinets with shallow drawers (intended for storage of stationery, cassettes, print-wheels, etc.) in office supply stores. Lane Geology/Paleontology cases work well, although the drawers are not suspended. Global Equipment sells cabinets ("LION High-Density Drawer Storage") with ball-bearing-suspended drawers that appear absolutely ideal for an element collection. The Lane case gives you more storage for the dollar, but the smaller, suspended drawers of the LION case would be more convenient. An element collection gets a lot of use, with constant opening and closing of the drawers.

To keep bones from rolling around every time a drawer is opened or closed, line the bottoms of the drawers, or the trays in the drawers, with velvet--not felt, it catches on everything.

Most of the bones in the element collection will be stored in open trays, but some minor elements are bagged in small zip-locks with an identifying stick-on label and stored in open boxes, rather like miniature file folders. Vertebrae are strung and hung on a wall.

You will need drawers/trays for the following categories:

skulls	furculae	fused pelves
mandibles	R & L scapulae	unfused synsacra
prefrontals	R & L coracoids	R & L unfused innominates
supraorbitalia	R & L humeri	unfused pubes
nuchales	R & L ulnae	R & L femora
vomers	R & L radii	patellae
palatines	R & L radiales	R & L tibiotarsi
pterygoids	R & L ulnares	R & L fibulae
R & L quadrates	R & L carpometacarpi	intertarsal sesamoids
syringes	R & L manual phalanges: I-1	R & L tarsometatarsi
atlases	R & L " " II-1	R & L metatarsal I's
axes	R & L " " II-2	R & L basal phalanges: I-1
notaria	R & L " " III-1	R & L " " II-1
pygostyles		R & L " " III-1
sterna		R & L " " IV-1

The following are stored in zip-locks:

sclerotics
hyoid/larynx
tracheae
caudal vertebrae
vertebral ribs
sternal ribs
pedal phalanges
ungular phalanges
miscellaneous (mostly sesamoids)

Vertebrae are hung by their tags on wire brads nailed into a strip of wood attached to the wall. I first nailed 3/4-inch wire brads (at a slight upward angle) one inch apart along the middle of the broad edges of three lengths of finished 1x2 pine. The length of the boards is determined by the amount of wall space you can free up (a hallway is a likely spot). A screw-in eye-bolt was mounted near the end of each board on its upper edge. The boards were hung from the wall with a picture hanger for each eye-bolt, at heights of 36", 57", and 73" above the floor. The vertebrae are hung by the holes in the key-tags, taxonomically from left to right, but with longer columns along the lower rows. Aesthetically, it's a nice touch to have measured the same amount of slack on each piece of string, so the columns appear to hang evenly. The wall-space where the vertebrae are hung should be well-lit. It's surprising how easy it is to identify vertebrae when they are organized this way.

Most of the pedal phalanges in the element collection are stored in 2x3 zip-locks, but I have found it convenient to put the phalanges of the more common larger taxa in "nut cups." These are tiny paper or plastic cups available in two sizes at party goods stores and some "dime" stores (e.g., Ben Franklin). The small nut cups are stored in "tart tins" (muffin tins with tiny compartments) and the larger ones are stored in standard muffin tins. Each tin holds a dozen cups, and I have found two large tins and five smaller ones adequate.

One of the smaller tins is reserved for holding the basal phalanges of representatives of the commonest families, not by taxon, but by element. The basal row of pedal phalanges is highly identifiable, and this is not surprising considering that they are associated with the distal end of the tarsometatarsus, one of the most distinctive and identifiable of avian skeletal elements. Because of their density, pedal phalanges are very common as fossils and subfossils, so it's worth setting up a system allowing their identification (e.g., the only specimen of Balaeniceps rex in the collection of Pliocene fossils from Hadar, Ethiopia, is of the proximal end of basal phalanx R-III-1).

As an element collection grows, it soon becomes efficient to break the collection down by size range. In practice, I have arrived at five size

classes: 1) most passerines and the smallest non-passerines, 2) large passerines and small non-passerines (up to teal-size, includes most of the charadriiforms), 3) medium-sized non-passerines (duck to cormorant size), 4) most larger birds (eagles, swans, etc.), 5) the longer bones of tall birds (cranes, storks, flamingos, etc.).

Given their distinctive size, geographic separation, and the few number of species involved, the larger ratites are not needed in an element collection for identification purposes. However, for teaching it is very valuable to have an ostrich, emu, and rhea organized by element. The three require a large case to themselves.

As the number of taxa represented in your element collection increases, you will want to make three taxonomic spin-offs: for anseriforms, charadriiforms, and passerines. I have found it practical to make a permanent geographic split in the collection: New World vs. Old World. Each is housed in separate cabinets, which means there are in fact two element collections. Minor adjustments in contained taxa are made in each as projects change.

CONCLUSION

I mentioned in the introduction that I strive for a method of preparing skeletons that produces the cleanest possible specimen with the least investment in person-hours, and I believe I have evolved such a method. In contrast, my method of labeling and organizing skeletons undoubtedly involves more labor than anyone else now commits to osteological preparations. I can only say that this effort, especially that invested in building an element collection, is definitely worth it in terms of efficiency in research. It's a big investment with an enormous long-term pay-off.

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PRODUCTS, VENDORS

steel shelving for archive boxes, parts bins for dermestid colony: Arrow Star Discount, 668 William St., Lynbrook, NY 11563 (also Elmhurst, IL and Carson, CA); customer service: 1-800-645-2982 (NY 516-481-1414); request mail-order catalog; variety of useful industrial equipment/furnishings

4-ply string cones: most any large office supply store; approx. $12.00

twine holder: Stock #GO4-X105, $5.99; Viking Office Products, 13809 Figueroa St., Los Angeles, CA 90061-0144 (also Dallas, Cincinnati, Windsor CT); 1-800-421-1222; request "office products buyers guide" mail-order catalog

Treasury Merital Cotton Non-Sterile: Treasury Drug Division, J.C. Penney Co., Inc., New York, NY 10019 (retail "Treasury" drug stores, usually adjacent to a "Penney's"); about $2 per .975-pound roll (sterile cotton rolls from Fisher Scientific and other scientific supply houses and drug stores are $10 and up)

Curity Cheesecloth, grades 10 to 80: The Kendall Co., 20 Walnut St., Wellesley Hills, MA 02181 (usually available through your local fabric store); about $.50/yd. by the yard (comes in 80-yd. boxes, usually at a discount); cheesecloth is also available from Fisher Scientific

small zip-locks: Grieger's, Inc., 900 S. Arroyo Parkway, Pasadena, CA 91109; 800-423-4181 (CA 800-362-7708); also lapidary supplies and other useful items, request mail-order catalog

ultrasonic cleaner: Cole-Parmer Instrument Co., 7425 N. Oak Park Ave., Chicago, IL 60648; 1-800-323-4340 (in Illinois, collect 312-647-7600); also small zip-locks and a myriad or other scientific products, request mail-order catalog

Terg-A-Zyme: Alconox, Inc., 215 Park Ave., So., New York, NY 10003, 212-473-1300; available through dealers (e.g., Curtin Matheson Scientific, Inc., 7524 Currency Dr., Orlando, FL 32809, 1-800-432-0324); about $12 for a 4-lb. carton

Geology/Paleontology Cases: Lane Science Equipment Corp., 105 Chambers St., New York, NY 10007, 212-563-0663; case is Model #301 (29"w x 32"d x 37"h) with room for up to sixteen 2-inch drawers, $340-370; drawers are Model #301-T (I.D. 24"w x 30"d), $32-35 each; base is Model #301-B, $32-35; prices depend on distance shipped; total price about $800-$900

LION High-Density Drawer Storage: Global Equipment Co., 6644 Hemlock Dr., Hempstead, NY 11550; customer service: 1-800-645-2988; Model 182108 (30"w x 28"d x 44"h) has eleven 3-inch drawers and one 5-inch drawer, (I.D. 25"w x 25"d), $951; drawer dividers available; request "industrial products buyers guide" mail-order catalog; variety of useful industrial furnishings and equipment

Deo-V deodorizer: Zep Manufacturing Co., P.O. Box 2015, Atlanta, GA 30301, 404-352-1680; contact for name of local distributor

TECHNIQUES FOR PREPARING BIRD EGGS AND NESTS

Lloyd F. Kiff
Los Angles County Museum of Natural History
and
Western Foundation of Vertebrate Zoology

INTRODUCTION

Bird eggs are used in a broader array of biological disciplines than any other type of avian specimen. Researchers in such divergent fields as toxicology, ecology, systematics, and anthropology have found museum egg collections to be a rich source of ready made data. In contrast, bird nests have been used as raw materials by surprisingly few researchers, and they represent a largely untapped source of potential new directions for research.

Formerly, the interest in collecting and describing bird eggs and nests was largely by hobbyists, and most natural history enthusiasts learned how to prepare eggs in their childhood. As egg collecting fell into disfavor with professional ornithologists and conservationists in the 1940s and 1950s, fewer young people learned how to prepare eggs as a rite of passage. With the renewed interest in egg collections as legitimate research materials, it has become clear that information on preparation techniques would be useful to a generation not exposed to traditional egg collecting. This summary is intended to cover only aspects of egg preparation. Details of data recording, shipping methods, and curatorial techniques for egg collections will be presented in a future report.

EGGS

General Considerations

As is the case with all types of natural history specimens, standard preparation methods yield eggshell specimens with the most comparative value and with the broadest range of research applications. The preparation methods outlined here are time-honored ones, for the most part, and they will result in specimens which can be compared directly with others over 150 years old. Indeed, the fact that eggshells of the latter age even exist and that they are at least superficially identical to specimens taken in the last decade can be taken as a verification of the soundness of the method.

A museum eggshell specimen is a largely inert object composed of calcium carbonate crystals arranged on a protein matrix. All or most of the eggshell membranes usually remain

attached to museum specimens. Only eggshells that are completely clean and empty have a potential for lasting indefinitely. Those containing residual conglomerations of yolk or albumen generally crumble within a few years, perhaps as the results of enzymatic reactions from the yolk. Unblown eggs invariably decay and often explode from the pressure of internal gases resulting from the processes of decomposition of their contents. Museum specimens are conventionally blown with a single blowhole, which leaves both ends and three sides of the eggshell intact for study or photographic purposes.

Unblown eggs are far more fragile than empty eggshells. Empty eggshells will break, however, if they come into rough contact with any object harder than themselves. Therefore, it is advisable to never pick up an egg specimen unless there is some good reason to do so.

Preparation Tools

Blowpipe: The type used in general chemistry labs, or a hypodermic syringe attached to a piece of rubber tubing.
Metal drills or finishing burrs: These are the sorts of drills used by dentists. The flame-tipped varieties are best. Those sold by biological supply houses are very inferior. Ideally, the drills should be cleaned with an ultrasonic cleaner between uses and coated with WD-40 while in storage, since they tend to rust badly.
Plastic bowl: Eight inches in diameter or larger.
Wash bottle
Paper towels
Thin stiff wire hook: This can be made from a bent insect pin.
Marking pen: "Crowquill" type for smaller eggs; fine-pointed "Rapidograph" type for larger eggs.
Permanent black (India) ink

Preparation Procedure for Fresh Eggs

Hold the egg between the thumb, forefinger, and middle finger of the left hand (right hand, if left-handed) over a plastic bowlful of water at all times during the blowing process. Eggs often become very slippery during preparation, and they are much less likely to break if they fall into a container of water than onto a work table.

With the free hand make a small puncture in the eggshell near the center of the egg with the sharp point of a small drill. The hole should be made on the side of the egg with the fewest markings.

Place the point of a small drill in the puncture hole and, by hand, rotate the drill slowly in a clockwise direction, taking care not to release it. On small eggs do not push the drill, as this may cause the side of the egg to collapse. A certain amount of pushing force is necessary to drill through the shells of larger eggs.

After the drill cuts completely through the shell, the ragged edges of the eggshell membrane should be cut away by rotating the drill lightly in both directions just under the inner lip of the blowhole. Otherwise, the projecting edges of the membrane will impede the free flow of the contents out of the egg. (A flame-tipped drill is ideal for this purpose).

After a clean round hole has been made, hold the egg so that the hole is downward. The tip of the blowpipe should be held a few millimeters away from the hole -- the blowpipe does not enter the hole. The egg contents are removed by alternately forcing jets of air into the egg through the blowhole, then allowing the egg contents to drain out of the same hole.

Air may be introduced by mouth, or by a squeeze bulb on the end of the blowpipe. The former method is preferable for small eggs, since it allows one to gauge the amount of air entering the egg more easily (too much pressure can easily cause a small egg to explode). The squeeze bulb technique can be used safely with large eggs, and it saves a good deal of wear and tear on one's cheeks.

Often, a bead of liquid initially forms at the blowhole, and the flow of this material (generally albumen) can be enhanced by stroking one's thumb across the blowhole. It is inadvisable and unnecessary to attempt to pull liquid contents directly out of the hole.

After the contents have been completely removed (which can be confirmed by flotation, determining if the egg rolls to one side when placed on a flat surface, or by "candling" the eggshell against a strong light), the empty eggshell should be rinsed repeatedly with clean water. Water can be introduced into the egg by using a water bottle equipped with a nozzle, or by sucking up water in the blowpipe and forcing it through the hole.

Place freshly-blown eggs hole downward on a blotter or paper towel to dry. The blotting surface should be inside a shallow container, since the slightest draft may blow an empty eggshell off a work table. In warm climates or in field situations eggshells should be protected from flies while they are drying.

Notes on Hole Size

The size of the blowhole should depend upon how far incubation has progressed. Incubation stage can be roughly determined by placing the egg in a container of water -- fresh or slightly incubated eggs sink, while heavily incubated eggs float. Alternatively, clear albumen generally first appears in the blowhole of fresh or slight incubated eggs, whereas the appearance of a dark yellowish yolk at a newly made hole is an ominous sign, since it usually signifies a well developed embryo. All fresh eggs, regardless of size, can be blown successfully with a hole 1 mm in diameter. Eggs containing developing embryos must be blown with proportionately larger holes. If a larger hole is necessary, it is safest to make the hole progressively larger by using a graduated series of drills. If the edge of the hole becomes chipped, it is exceedingly difficult to prevent further extensive fracturing of the shell. Do not try to "round out" a chipped hole, as this is usually impossible.

It is important to use a small blowhole, if possible, since eggs become progressively weaker as the hole is made larger, and specimens with very large holes are less useful to researchers wishing to obtain eggshell weights. Never sacrifice a specimen, however, by attempting to remove the contents through too small a hole.

Procedures for Removing Large Embryos

Small and intermediate-sized embryos can be removed from eggshells without much

difficulty, and even very well developed embryos can eventually be extracted with patience. Embryos larger than the diameter of the blowhole must be removed from the egg piece by piece. This means that the embryo must be disarticulated while in the egg, so that the various appendages can exit end first through the blowhole. The ultimate limitation to minimum blowhole size is probably the diameter of the largest hard bone in the embryo.

After first removing as much of the liquid component as possible, introduce water into the egg. When it is nearly (but not completely) refilled, the egg should be placed in the upturned palm of one hand (beware of rings!) with the fingers closely tightly over it. While holding the specimen in this manner, pound the wrist briskly with the other fist. This produces a turbulent confrontation between the egg contents and the water which was introduced into the egg, resulting in the gradual disarticulation of the soft-bodied embryo. Repetition of this process, alternating with blowing, will generally result in the removal of all but the largest embryos.

Very large embryos may be removed from eggs by the use of pepsin, a protein-digesting enzyme. Formerly sold under the brand name "Caroid", a suitable source of pepsin is now papaya extract, or even commercial meat tenderizers. An aqueous solution of the enzyme should be injected through the blowhole into the actual embryo using a hypodermic syringe. Pepsin will not damage the calcium carbonate portion of the eggshell, but it will break down the eggshell membranes and also any superficial pigments on the eggshell surface. For this reason, it is important to avoid letting any of the enzymatic solution fall onto the surface of the egg.

An egg injected with pepsin should be filled as full as possible and placed in a container with the hole upward. At daily intervals an attempt should be made to blow the egg again, and any decomposed material should be extracted. The enzyme works gradually, so the repeated blowing process will remove a little more of the embryo each time. Eggs with embryos near hatching may require two or three weeks to clean in this fashion. During this period it is important to ensure that the egg contents remain moist to prevent the contents from solidifying and stimulating a process of shell decomposition. I have not yet found a satisfactory method for removing the dried contents of eggs without damaging the shells.

The most difficult portions of any large embryo to remove through the blowhole are the pectoral and pelvic girdles, especially if they have developed beyond a soft cartilaginous stage. In extreme cases, such bony structures can be teased out of the blowhole by the use of a thin, stiff wire hook. This is a risky process, and it may result in a chipped hole. Other than this specific situation, or when injecting embryos with a pepsin solution, I do not recommend placing any instrument inside of the eggshell during the preparation procedure.

Marking

All eggshell specimens should be inscribed with some distinctive notation so that they can be identified in case they become separated from others in the set or from the original data. Unmarked eggs easily become mixed up with other unmarked specimens, rendering them all scientifically useless.

Eggs should be marked with permanent black ink (or, in the case of extremely dark-

colored eggs, with permanent white ink). Pencil marks are difficult to read and less permanent, and "magic markers" or felt tip pens tend to produce messy, overlarge marks. A "crowquill" pen point, the smallest that can be obtained, is ideal for marking passerine eggs, no matter how small the specimen. "Rapidograph"-type pens are useful for marking larger specimens, e.g., crow egg-sized or larger.

Egg marks should be as small and neat as possible, so as to interfere with the least amount of egg surface. The marks should be unique for each "set", or clutch of eggs, and they should always be made around the blowhole. It is best to avoid such traditional notations as "1/4" or "2/5" since they are too non-specific. Most large museum collections contain many presently unidentifiable eggs with such generic markings.

Other than providing some sort of unique tag, there is no "correct" way to mark eggs. Among the types of information which can be marked on the egg are the following:

Catalog number: One's own catalog number. Chronological numbering systems are probably the best for this purpose, as they insure that each set will have a unique mark.

Number of eggs in the set: This is often recorded beneath a slash mark with the catalog number on top.

Date: Year only, or the whole date. In the latter, case, it is advisable to indicate the month with a Roman numeral because of the perennial confusion between Old and New World notations.

AOU number: This is available only for those forms treated by the first five editions of the "AOU Check-list of North American Birds", but has traditionally been a standard portion of the marks on the eggs of these species. The AOU number provides a quick means of identifying specimens to species.

Personal mark: This may take the form of an initial or other symbol, which is marked on all the eggs taken by a particular collector.

Egg number: If whole egg weights are taken, or if the laying sequence is known, it is useful to identify individual eggs in the set by number. This may be done by placing the egg number immediately beneath the blowhole.

NESTS

General Considerations

Bird nests come in such a diversity of forms and sizes that it is not surprising that few comprehensive nest collections are maintained. Although most natural history museums cannot spare the space to store bird nests in large series, it is useful to maintain at least a basic reference collection for local species. Given care in handling and protection from dust, such specimens can retain a relatively fresh appearance for decades.

Preparation and Collecting Tools

Tree clippers
Handsaw

Toilet paper or paper towels
Thread
Aluminum foil

Preparation Procedures

The procedures described here are mainly applicable to the typical cup-shaped or pensile nests of passerines and some families of non-passerines. Ground nests, burrow nests, and cavity nests all present special problems which cannot be adequately discussed in this brief summary.

It is useful to decide at the outset whether the nest is being collected for display or research purposes. Display specimens should be accompanied by a relatively large portion of the substrate, whereas specimens taken for research use should as compact as possible without a loss of biologically significant information. Regardless of their intended use, however, all nests should be collected in a manner that preserves the point of their attachment to the substrate, as practical. Tree nests should be collected with a piece of the branch supporting them, i.e., they should not be plucked bodily from the supporting limb.

A padding of paper towels or toilet paper should be placed in the nest cup to retain its shape during transport from the field. The nest should then be wrapped fairly lightly in toilet paper or paper towels. The intent of this procedure is to "capture" all the loose ends of the nest and to make it less vulnerable to damage during handling. The nest should then be wrapped with thread, but not so tightly as to disrupt the natural shape. Finally, the whole affair should be wrapped very loosely with aluminum foil. The resulting compact package can then be readily transported from the field in a sturdy cardboard carton. Fresh nests packed and transported in this manner should be unpacked as soon as possible and restored to their original shape.

I recommend strongly against the use of cotton anywhere in the vicinity of bird nests, since it tends to leaves wisps which are almost impossible to remove later. Since some bird species actually use cotton in nest construction, the remnants of cotton packing material can leave a misleading impression with later collection users. Recently, commercially available plastic netting has been wrapped by some researchers around nests to hold them together in shipping. While this is effective in preserving the nest shape during transport, the later removal of the netting can be quite tedious and cause serious damage to the specimen. Another deleterious procedure recommended in some older texts is to apply shellac or some similar coating to the nests to preserve their structural integrity. We have not found this to be necessary, and such coating results in an artificial appearance and can obscure structural details.

A field label should be attached to the nest at the time of collecting, and this should ideally be sewed right through the nest wall.

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