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OBTAINING, PRESERVING, AND PREPARING BIRD SPECIMENS

KEVIN WINKER
University of Alaska Museum
907 Yukon Drive
Fairbanks, Alaska 99775 USA

Abstract.—The scientific value of avian research specimens is immense, but the accumulation rate of this resource is too low to meet either present or future needs. This may be due, in part, to the fact that few students are currently being taught to prepare specimens. Modern specimen preparation is a routine but detailed and meticulous process in which comparatively few are expert. I summarize methods for obtaining bird specimens and preserving them both for the short term and for the long term as high quality scientific research specimens. The preparation method outlined preserves skin, partial skeleton, stomach contents and two duplicate tissue samples for every specimen, maximizing the scientific usefulness of each bird. The resulting skins and skeletons augment current samples, simultaneously increasing the sample sizes available for studies involving either type of specimen. These methods allow a diverse array of data to be taken from every individual, and are thus suitable for general preparation or focused, single-species research projects. These archival quality methods assure that, if prepared as outlined, the skin and skeleton specimens possess a useful life of half a millennium or more. I suggest that this is an unparalleled opportunity to make a personal, signed, long-term contribution to science with relatively little time investment.

OBTENIENDO, PRESERVANDO Y PREPARANDO ESPECÍMENES DE AVES

Sinopsis.—El valor científico de especímenes de aves para investigación es inmenso, pero la tasa de acumulación es muy baja para satisfacer las necesidades presentes o futuras. Esto se puede deber en parte al hecho de que pocos estudiantes están siendo entrenados a preparar especímenes. La preparación moderna de especímenes es un proceso rutinario pero detallado y meticuloso en que hay pocas personas expertos. Resumo los métodos para obtener especímenes de aves y preservarlos tanto en corto tiempo como para especímenes de investigación de gran calidad científica a largo tiempo. El método de preparación descrito preserva la piel, esqueleto parcial, contenido estomacal y dos muestras de tejido para cada especímen, maximizando la utilidad científica de cada ave. Las pieles y esqueletos resultantes aumentaron las muestras presentes, aumentando simultáneamente los tamaños de muestras disponibles para estudios requiriendo cualquier tipo de especímen. Estos métodos permiten una obtener una diversidad de datos de cada individuo, y por lo tanto son apropiados para la preparación general o enfocada, y de proyectos de una sola especie. Estos métodos de calidad de archivo aseguran que, de prepararse tal como se describe, los especímenes de piel y esqueletales poseen una vida útil de al menos 500 años. Sugiero que esta es una oportunidad sin paralelos para producir una contribución personal de larga duración firmada a la ciencia con una inversión de tiempo relativamente corta.

The importance of specimens to the science of ornithology and the conservation of biodiversity would be difficult to overstate (see Remsen 1995; Winker 1996, 1997). Yet, at a time when the need for avian specimens has increased across a broad range of studies, it seems that few are being added to collections, even though the world's bird collections in aggregate represent a grossly inadequate documentation of extant birds (e.g., Goodman and Lanyon 1994, Winker 1996). This may be due in part to the fact that few students are being trained to prepare specimens (cf. Rogers and Wood 1989). Given the state of literature on the subject

(mostly outdated) and the changing nature of specimen preservation, even a motivated novice would have a difficult time learning to produce high quality research specimens. While still containing some useful pointers on various aspects of specimen acquisition and preparation, earlier guides to specimen preparation (e.g., Swainson 1836, Coues 1874, Ridgway 1891, Chapin 1946, Blake 1949, Hall 1962, Proctor and Lynch 1993) are outdated and not sufficiently comprehensive. Johnson et al. (1984) provided an excellent contribution emphasizing tissue preservation and updating older preparation methods with important new variations, but was neither comprehensive nor of sufficient detail for the novice.

With a little foresight, understanding, and practice, field ornithologists can preserve the specimen research material that is needed today and that will be highly useful in the coming centuries. Because a specimen is extremely useful for many types of research, and because proper preparations can last for centuries, it behooves all ornithologists to promote the proper collection, preparation, and preservation of bird specimens. It should be remembered that the populations these specimens document represent renewable resources.

Here I outline practical methods for obtaining, preparing, and preserving bird specimens that maximize the usefulness of each bird. These methods are intended to be useful to the novice for learning to prepare and preserve scientific bird specimens, but also to the professional, who might find new ideas and methods that increase the research value of each specimen. The methods outlined here are for general purpose specimens, and are therefore not all-encompassing. Although more intricate than many previous methods, these methods are not overly cumbersome, given the long-term scientific value of the products, and they have proven suitable for general preparation procedures. These methods are also suitable for focused research projects examining broad suites of morphological and genetic characters.

PHILOSOPHICAL APPROACH

Today, specimen-based ornithologists practice a wide range of preparation procedures (e.g., Johnson et al. 1984). Many of these methods are specialized for the immediate needs of the researcher, and workers often needlessly discard material desperately needed in the broader scientific community simply because it is too much time or trouble to properly preserve portions of the specimen they don't use. This situation applies even to some dedicated collectors, who will throw away skin, skeleton, or tissues because they personally don't use that particular type of bird sample. As a general methodology this is difficult to justify. The skin, skeleton, and tissues of every bird can and should be preserved whenever time allows.

At the root of the method presented here is a ''total evidence'' approach: the more evidence that we are able to bring to bear on a question, the more confidence we can have in the answer or results derived from the specimen material. This suits not only individual research projects,

but also brings to the present and future research community most of the components of a dead bird that we know can be useful, whether for traditional studies of phenotype and genotype, or for stable isotope and contaminant analyses, or for future methods (and questions) as yet undeveloped. From the specimen use requests I have seen in the past few years, it is certain that museum collections will continue to provide an increasingly broad research community with immensely valuable historic research material for an amazing variety of questions. Preserving specimens with this future in mind significantly enhances both our science and our personal contributions to it—particularly at a time widely viewed as a watershed period of biodiversity loss and climate change.

The preparation method presented here emphasizes a combination skin-skeleton specimen that enables the skins to enhance existing samples of skin collections. Thus, skins without bills (often called ''shmoos'' or ''muppets'') are de-emphasized, because these remove the important ramphotheca from the skin, making these skins incomparable (or incompletely comparable) to previously existing specimens. Consequently, the skeletons produced using this method are partial, in that the full skull is not preserved. Although there are many differences, this method is most similar to Method 2 outlined by Johnson et al. (1984).

Preserving both skin and skeleton from each individual whenever possible is fully justified. The need for skeletal material has been emphasized for at least 140 yr (Newton 1860), and the now outdated world inventories of skeletal specimens (Wood et al. 1982, Wood and Schnell 1986) had a profound influence on complete skeleton preparations. But plumage probably reveals more of the developmental similarities and differences among populations than skeletons, and is also useful for examining age-related differences among individuals. Although we are too ignorant at present to use and decipher all of the information available in plumage characteristics, tools such as reflectance spectrophotometry are providing increased resolution and rigor when studying plumage characteristics (e.g., Graves 1997). Skeletal characters are useful at multiple levels and are not subject to the fading and wear that plague plumage studies. Furthermore, series of skeletons are as important as series of skins for incorporating individual variation into data sets, and sample sizes of both skins and skeletons are maximized by preserving both from each specimen. Finally, and importantly for focused projects, studies of evolution are improved when broad suites of genetic and morphological characteristics can be assessed from the same individuals (see also Johnson et al. 1984).

What should be preserved and prepared?—The world's systematics collections are so weak in so many areas, and so few specimens exist to document today's populations, that almost every dead bird has scientific value. In ongoing morphometric studies I have found it difficult or impossible to assemble 30 individuals of each age and sex class of many common eastern North American passerines. This stems in part from the fact that older specimens have fewer data than modern preparations (most lack

data on degree of cranial ossification and body mass, for example), but it is also due to a general paucity of specimens.

As members of a research community concerned with increasing our knowledge of birds in the face of accelerating global changes, we will achieve much greater success by pulling together than by blindly pursuing only our own narrow research agendas. A certain degree of altruism benefits us all, and, importantly, provides immense benefits to future researchers. If preceding generations of ornithologists had not made general collections and preserved them, our science would be greatly impoverished today. Whenever we go into the field we might be the only trained ornithologists to visit an area in decades. Consequently, anyone in the field should be supported and encouraged to sample broadly: few areas are adequately sampled, and temporally adequate samples essentially do not exist. Depositing specimens in research collections that have wide accessibility is an important corollary of this philosophy.

With the growth of the animal rights movement has come an increasingly strident opposition to the active collection of birds. Personal beliefs in this regard are to be respected, but I have argued elsewhere that in many cases this opposition is misguided, often arising from an ignorance of biological principles and the value of the specimens themselves (Winker 1996). Here I wish to emphasize that one's beliefs about how animals should be treated (i.e., allowed to die) should have little or no effect upon one's dedication to preserving specimens for science. The avian carnage caused by domestic cats, glass windows, automobiles, and communications towers is far greater than the comparatively tiny numbers of birds collected by scientists each year (Banks 1979, Churcher and Lawton 1989, Klem 1990b). It seems a terrible breach of ethics to allow this carnage to go to waste when the specimens are often of great scientific value. And, in fact, probably most specimens added to museum collections today come from the salvage of birds found dead. A good salvaged specimen is one less that might have to be collected. Thus, individuals ethically opposed to the collection of birds might be looked upon to become some of the best and most active preparators, wishing to see something beneficial resulting from the many avian casualties arising from nonscientific anthropogenic influences.

The bird bander represents an important category of avian researcher whose activities provide unique opportunities to obtain and preserve research specimens. Unfortunately, these opportunities are rarely exploited. Although the work of banders has benefitted enormously from information derived from bird collections, and though banding regularly generates dead birds through accident, there is little return to collections from banding efforts. I am a bird bander, and have learned through experience that the quality of my work is greatly improved through a combination of banding and selective collecting. If a bander is really motivated to learn about birds, rather than simply deriving enjoyment from capturing and banding them, then it is a very short step to discovering how much more can be learned through judicious collection.

I regularly use skins 100–150 yr old in my skin-based research. Although the purposes for which I am using them are probably different from the reasons for their initial collection, their scientific and physical integrity remain as great or greater than the day they were prepared. In fact, their scientific value often increases with time through label annotations and biotic changes occurring among populations and environments. Judging from old books, which are also products of plant fiber and animal skin, we can realistically expect the archival-quality skin preparations outlined here to last for at least 500 yr or more if housed under proper conditions. I can think of no other comparatively short term time investment with such staggering long term scientific impact.

PERMITS

The collection and possession of bird specimens is generally regulated, and many types of permits are often required, depending on the country or countries where these activities take place. The long-term scientific value of a specimen is dependent upon its being accessible to the scientific community. The scientific community abides by all permitting requirements, and therefore requires that specimens be legally acquired and possessed. As scientists, it is our duty to obtain permits that enable us to perform our jobs to the best of our abilities, but which at the same time provide adequate safeguards to ensure that wildlife resources remain available for continued existence and for use and enjoyment by society at large. There have been conflicts between scientists and permitting agencies in obtaining permits agreeable to both parties (for some discussion, see Remsen 1995, Winker 1996). However, it is usually possible to work with permitting agencies to achieve mutually agreeable permits. Make every attempt to provide permit-granting agencies with the information they need to make informed decisions. In general, scientific collecting does no harm to avian populations (see Banks 1979, Remsen 1995, Winker et al. 1991, Winker 1996). If one wishes only to salvage dead birds, special salvage permits are usually issued, and are generally easier to obtain than collecting permits.

An important category of permit is that allowing the international import and export of specimens. In some countries the collecting permit also serves as a valid export permit, while in others two separate permits are needed. In the U.S., a national U.S. Fish & Wildlife Service (USFWS) import-export permit is required for any international transaction, and, in the case of import, an export permit is usually needed from the country of origin. In the U.S., specimen import is also regulated by the U.S. Department of Agriculture's Animal and Plant Health Inspection Service (USDA-APHIS; www.aphis.usda.gov). Thus, in the U.S., permits to transport and possess wild bird specimens should be obtained from both USFWS and USDA. Learning of the specific requirements prior to any field collecting or international transaction can save much time and trouble. Realistically, plan for permitting processes to take 6–12 mo before field work can be conducted. And be aware that many different permits

might be required, depending on where your activities are being performed. For a recent collecting trip to Canada, for example, I needed a total of seven permits.

Specimens of species listed under national laws concerning threatened or endangered species, and species listed in the Convention on International Trade in Endangered Species (CITES) appendices usually require special permit procedures. Many international specimen shipments require CITES permits (sometimes even if the taxa involved are not CITES-listed!).

In some countries, researchers at institutions must obtain permission from Institutional Animal Care and Use Committees (IACUC, or an equivalent) to study birds using the methods outlined by the researcher in a proposal to the committee. The duty of these committees is to assure the ethical and humane treatment of animals in science, and workers adhering to professional guidelines (Gaunt and Oring 1997) should have little difficulty in obtaining IACUC approval. Some funding agencies (e.g., in the U.S., the National Institutes of Health and the National Science Foundation) will not make grant awards available until IACUC approval is obtained. Although it is not within their purview, some IACUC committees do not approve of the scientific collecting of birds, and it may be difficult to convince them of the need to actively collect specimens. In these cases and in permit applications it is often useful to point out the scientific benefits and justifications for collecting birds, and that the numbers taken are small in relation to natural and other human-related mortality factors. Publications where these and other useful data and arguments have been assembled include Banks (1979), Winker et al. (1991), Goodman and Lanyon (1994), Remsen (1995), Winker et al. (1996), and Winker (1996, 1997). Educating permit-granting agencies, committees, and personnel, while at times rewarding, has become a time consuming and often frustrating part of the business.

Permitting procedures change frequently, and I know of no better method to proceed than writing or telephoning the appropriate agencies to request a set of current guidelines. Association of Systematics Collections (1993), searches on the internet, and inquiries to biologists familiar with local permitting conditions can speed the process.

IN THE FIELD

Methods of obtaining birds are too varied to be treated in great detail, but a brief summary is useful for the novice. Most birds added to research collections today probably come from the salvage of birds found dead, but collecting birds using shotguns or traps remains an important means of acquiring specimens not obtainable through salvage. Bub (1991) gave methods of trapping; the mist net is probably the most commonly used trap in general use today (see Appendix for addresses of suppliers).

Collecting birds using firearms has become an arcane aspect of field ornithology, despite its historic prominence. For larger birds, the standard firearms and ammunition used in game bird sport hunting are effective.

For smaller birds, however, specialized equipment is necessary. Museum collecting has become centered on double-barrelled shotguns, usually using either 12, 16, or 20 gauge weapons with auxiliary barrels. Auxiliary barrels are machined metal inserts that enable one to fire a smaller caliber shell from a weapon chambered for something larger. For example, auxiliary barrels can enable one to fire .410 or .22 caliber shotshells from a 12, 16, or 20 gauge shotgun. ''Aux'' barrels are easiest to use when less than about seven inches long. They drop into the shotgun chamber like a long shell, and they are removed for reloading or when one wishes to use a full-sized shotgun shell. Carrying a 12-gauge, double-barrelled shotgun with .410 and .22 auxiliary barrels enables one to collect anything from geese or cranes to hummingbirds with the same firearm. The weapon is normally carried with a different caliber in each barrel to enable the proper load to be used when required, depending on species and distance (e.g., .410 and .22, or 12 gauge and .410). Firearms safety and laws regarding firearms are as applicable to bird collection as to any other use of firearms. Always assume the gun is loaded and be sure it is pointed in a safe direction.

When collecting, one wishes to dispatch a bird quickly and cleanly while at the same time minimizing damage to the specimen. Here, shot size and distance are the two most important considerations. Proper distance can be gauged through experience and practice, and varies with load and firearm. Most museum collecting of smaller birds using the smaller shotshells (.410 and .22) is done with No. 12 lead shot. No. 12 shot is only available in lead, and usually must be specially ordered. Loads containing shot of this size are commercially available in the U.S. in .22 caliber shotshells, but not in .410. For the latter, most active North American collectors are probably now reloading their own custom ammunition. Larger shot sizes (No. 9 is available commercially in .410, for example) are appropriate for larger birds (e.g., jays), but are not as effective as 12 shot for smaller birds because the pattern (spatial distribution of the shot) is not as dense.

Dispatching birds is probably the most difficult task faced by the collector—not physically, but psychologically. Generally, those of us studying birds do so because we have developed a strong liking for them. Consequently, actually killing them is decidedly unpleasant. However, having the specimen, which you know is valuable for the science of ornithology, is often a great pleasure, regardless of the specimen's source. The collecting of birds for science is unquestionably justified, and this justification keeps the collector active. ''But let all your justifiable destruction of birds be tempered with mercy; your humanity will be continually shocked with the havoc you work, and should never permit you to take life wantonly. Never shoot a bird you do not fully intend to preserve, or to utilize in some proper way.'' Coues (1874:30).

Two methods are generally used to dispatch birds in the hand, whether wounded or in traps: thoracic compression and cervical dislocation. The former is preferred in small birds because it is simple, quick, and causes

no damage to the specimen. As I practice and teach it, thoracic compression involves slipping thumb and forefinger in under the feathers on each side of the bird (positioned between the spine and sternum), softly feeling the ribs to be sure the position is correct, placing the forefinger of the other hand against the front of the sternum, and instantly bringing all three fingers together with a great deal of pressure focused on the heart and lungs. This instantly stops the heart and lungs, and must cause blood pressure to skyrocket. For small birds, it seems that unconsciousness occurs instantly; death follows very quickly. Pressure is held until it is certain that the heart won't restart. Twitches of the muscles can often be felt quite plainly; pressure should be maintained until they stop. This method is fast and, in my opinion, essentially painless. I get no enjoyment from this difficult task, and use the method because it is so quick and seemingly humane. I stress to students that anyone who might enjoy this should find another field of study.

Cervical dislocation involves quickly stretching the neck (not twisting) until the spinal cord is broken. Because thoracic compression is not sufficiently rapid in larger birds, cervical dislocation is preferred. The latter method is not usually acceptable in small birds because it can easily damage the specimen and makes skinning more difficult. Methods used by veterinarians to dispatch birds usually involve drugs or gasses that are often illegal, inconvenient to carry in the field, damaging to the specimen's value, or of some risk to personnel.

Preparation is made easier by preventing body fluids from getting onto the plumage. This process begins as soon as one picks up a bird. Living birds should be held with the cloaca pointed downward. Freshly shot birds, or any bird that is wet or leaking body fluids, can be placed in a bag of cob dust, sawdust, or corn meal to absorb moisture and keep the plumage clean. Also, a wad of absorbent cotton or tissue paper should be put down the throat—this is the most common source of fluids leaking onto the plumage. Perforations of the skin (e.g., from shot, cats, raptor talons, impact wounds, etc.) can also be plugged using cotton or absorbent paper to prevent leakage. Washing and drying feathers adds a lot of time to the preparation process, so preventing them from becoming dirty in the first place is worth the time invested here.

Specimens should be kept as cool as possible until they are prepared or temporarily preserved through freezing or fluid preservation. When external body fluids have become stabilized or dried in a bag of dust, I generally place the specimen into an open plastic bag kept or carried in shade away from my body until it can be labelled and frozen. Plastic bags may be used rather than the paper cones of yesteryear: while continuing to prevent plumage disarray they do not need to be made up in the field, and they do not contribute to subsequent desiccation of the frozen specimen. Every opportunity should be taken to allow birds to cool and to keep them cool until short- or long-term preservation is possible. A thin canvas bag, small pack in cool weather, or even a fishing creel or outer coat pocket can be used to carry dead birds in the field. Just remember

to allow the body heat to escape and to prevent feather disarray. Dead birds should always be handled in a manner that preserves the integrity of the feathers. Bending feathers and causing them to be ruffled backwards should always be avoided. One need not be particularly delicate; just be sure that contact with the bird goes with, rather than against, the grain of the feathers. Never grab or hold a bird by its tail or wings.

Freezing birds.—The best way to preserve birds when there is not time to prepare them immediately is to freeze them. Tissue decomposition begins immediately upon death, and substantial degradation of important elements can occur within the first hour. For example, the gonads of immature passerines can become unrecognizable within 1 h of death (even 30 min in warm conditions), and proteins also deteriorate, making it necessary to freeze birds (or a tissue sample) relatively quickly after death if they are to be useful for studies of allozyme genetic markers. Freezing birds within 0.5–2 h of death is best (cf. Johnson et al. 1984). Degradation of DNA also begins following death. Freezing or otherwise preserving tissue samples for DNA studies is highly desirable as soon as possible after death (see below). Specimens should be sexed when tissues are taken.

Often, freezing the whole bird is desirable. Preparation is usually easier in a lab than it is in the field, and, when it is possible to freeze specimens for later preparation, the return on the investment of field time can be maximized. If a freezer is not available, dry ice (solid CO_2) is preferred for temporarily freezing birds in the field (and also for shipping them). If kept well insulated, a 23-kg block of dry ice can last as long as two weeks in rather warm conditions, but this is exceptional and requires little use of the ice: few exposures to ambient temperatures and only a few small specimens frozen. Dry ice in blocks is far superior to pellets, pucks, or small blocks of condensate compressed straight from a CO_2 tank.

Whenever freezing a specimen collected or found dead, each bird should have a label containing data on date and locality (at least), and habitat and other notes as well, put into a plastic bag with it or tied to its leg prior to freezing. When using dry ice, place specimens directly on the ice in a cooler.

A standard chest-type cooler can be modified for improved use with dry ice by making a nested series of ''dense styrofoam'' boxes, with lids, to fit inside (J. Klicka, pers. comm.). Standard styrofoam is subject to severe deterioration under the temperatures of dry ice and liquid nitrogen; the blue kind of dense styrofoam is the best to use when exposures to dry ice or liquid nitrogen are expected. As the ice sublimates and shrinks, the cooler's contents are transferred to the next-smallest box, which is inserted into the cooler. This apparatus works quite well, but caution must be used to keep some ice on top of the specimens; I have had temperature differentials of over 45 C develop inside such a cooler under hot conditions.

A less common and more expensive method of freezing specimens in the field involves the use of liquid nitrogen (LN_2) in dewars, or flasks.

LN_2 is commonly used in the field to freeze tissue samples, and works very well for this purpose (Johnson et al. 1984). It works less well for the preservation of whole specimens, however. With care, it can be used successfully with small birds, but some specimen damage is to be expected. Upon immersion, liquid nitrogen seeps into the external air pockets of a specimen. When the specimen is removed from the liquid, the nitrogen expands very rapidly into a gas, causing explosive destruction of the walls of these air pockets. The rachides (shafts) of flight feathers (rectrices and remiges) are often riven, or shivered, as a consequence. Structural damage and even partial loss can occur in these feathers when specimens are frozen whole in liquid nitrogen. Less frequently, bills or other peripheral parts are damaged. This damage can be minimized by preventing rapid temperature change in the frozen specimens. You can actually hear the damage occurring as small pops and clicks upon rapid removal from LN_2. Holding specimens in the vapor phase of LN_2 for several hours or overnight diminishes damage considerably.

Using this method, specimens are not placed directly into LN_2; they are first wrapped. Plastic bags do not survive immersion in LN_2. Wrapping birds first in multiple layers of paper tissue (bathroom or facial tissues work well), then enveloping them in two layers of aluminum foil (label inside) works reasonably well (G. Graves, pers. comm.). These envelopes are then dropped into a wide-mouthed dewar for transport or placed inside the canes of small-mouthed dewars. This method is only usable with relatively small birds (ca. <60 g). When removed from LN_2, birds should be put directly into a -80 C freezer or onto dry ice pending preparation to maintain tissue quality. A final source of damage to LN_2-frozen specimens is that they are so cold that they are very brittle. An aluminum foil sarcophagus or envelope prevents most damage, but care must be taken to treat these specimens very gently. Do not drop them onto a hard surface.

For brief periods of freezing, ''blue ice'' (reusable plastic vessels filled with a refreezable fluid or gel) of at least -20 to -80 C can be used to bring birds from the field to the freezer. Although messier, bags of water frozen to -40 to -80 C also work if blue ice is not available.

Storing frozen specimens.—Freezing provides only temporary preservation; deterioration also occurs in frozen birds. Freeze-drying, the evaporation or sublimation of water out of the tissues, is the most common problem with frozen birds: water escapes from the carcass into the air surrounding it and recondenses outside the tissues. To minimize this problem, it is important to (1) minimize the time a bird is in the freezer; (2) minimize the amount of air that the specimen is exposed to (use plastic bags with as much air as possible removed from them); (3) maintain a temperature of -20 C or colder, if possible; and (4) minimize temperature fluctuations. Frost-free freezers are to be avoided at all costs. They undergo dramatic fluctuations in temperature designed to thaw, vaporize, and remove water condensation in the freezer compartment. This action mobilizes water in bird carcasses also, resulting in its recon-

densation in the bag, but outside of the tissues. Buffering specimens from temperature changes and exposure to air is important. Small specimens that have been triple-wrapped in plastic bags with all of the air possible removed from them, then placed inside styrofoam coolers in a constant temperature freezer, can be prepared quite easily up to three years following freezing. This is an exceptional length of time, however, and freeze-drying is usually detectable in the outer joints of small birds within the first year of freezing. Preparation of frozen specimens that have suffered freeze-drying can be assisted by soaking the affected parts (using soaked cotton) or even the whole bird in water in a refrigerator for a day or two.

Taking, preserving, and archiving tissue samples.—Specimens frozen whole on dry ice or LN_2 very quickly following death provide tissue samples of high quality for genetic studies. It is important to take tissue samples from these specimens as soon as possible to maintain this usefulness. Whole frozen specimens held at −80 C prior to preparation can be brought slowly up to room temperature by being held in an ordinary (−20 C) freezer for hours or days first, or brought directly to room temperature under close observation. Tissues should be taken and refrozen as soon as the specimen has thawed sufficiently to be worked. Usually, tissues can be taken while the organs are still frozen. With experience in preparation, in small birds the skin can be removed from the carcass and tissues taken before the internal organs have thawed. Two tubes of tissues should be preserved whenever possible. These should be stored separately, if possible, for both the short and long term to prevent loss of critical material through freezer failure or other catastrophe. Also, tissue tubes should be filled to within 2–3 mm of their tops whenever possible. This maximizes the amount of tissue available for future studies, minimizes wasted freezer space, and prevents tissue desiccation.

Taking tissue samples from each specimen should be a priority for anyone collecting regularly, and can be done by anyone who has the opportunity to work with dead birds. Winker et al. (1996) emphasized the crucial nature of voucher specimens when taking tissue samples. I will not discuss sampling protocols that do not preserve voucher material (e.g., bleeding and releasing live birds). Such cases should be exceptional. Traditional allozyme studies use several different tissues because of the different proteins available in each. In general, heart, liver, muscle, and kidney are saved for these studies (Johnson et al. 1984). Although allozyme studies have declined in popularity with the advent of improved DNA technologies, when one is freezing fresh tissue samples it is a good idea to preserve all of these tissues in each tissue tube. Doing so in the order above insures that the types are separable later in the molecular laboratory. For modern DNA studies, frozen tissues are not absolutely required, and freezing has become less popular because of the extra field logistics required (LN_2 or dry ice). This trend is short-sighted, however; frozen tissue samples are still preferable. When freezing tissues is not possible, one can place minced tissues in a vial of buffer (Seutin et al.

1991) or place small pieces into 95% ethanol for dehydration. New archival methods of taking and storing genetic samples will be or are being developed (e.g., using blood-soaked paper that has been chemically treated), but thus far ultracold freezing represents the closest thing to a museum-quality tissue preservation method. Given DNA chemistry, aqueous solutions do not represent long term, archival-quality storage conditions (see Cann et al. 1993, Poinar et al. 1996). Thus, aqueous field storage methods should not be trusted over the long term (years), and should be stored in cryosystems upon return to the museum or laboratory.

Labels and field catalogues.—Proper data recording and management is perhaps the most important aspect of specimen preservation, and doing it right begins in the field. Labelling is also where most errors occur. An improperly labelled specimen is of limited scientific value, and a specimen with no data may be useless. ''. . . never put away a bird unlabelled, not even for an hour; you may forget it, or die.'' (Coues 1874:70).

Historically, proper data management could be achieved using the label alone. This becomes much more difficult when several pieces of the same animal are preserved, however, because these pieces need to be easily linked when being archived and, eventually, used. The method outlined here is centered upon a field catalogue, and the link among these separate pieces is the unique field catalogue number. Consider that each preserved part is destined for a different physical collection (e.g., skin, skeleton, tissue), and that pieces often come to reside in different institutions. The field catalogue becomes the central data source for all of the preserved parts, because full labels will not be written for each. The field catalogue is never meant to be housed with a specimen, however, and time, together with specimen loans and transfers, assures that the field catalogue is often not available. Attached labels are therefore the most important and most used documents for data retrieval. My method uses the skin label as ''the final label;'' it will be as complete as the field catalogue entry and will be the only label with all of the data.

Personal field catalogues are a tradition in ornithology, but, as a consequence, many have been lost. Thus, beginning a personal catalogue should only be done when there is not another immediate cataloguing option. I regularly use project-specific and laboratory ''field'' catalogues to bring together under one system the efforts of many individuals who would otherwise generate an uncontrollable proliferation of idiosyncratic note taking. I have found a form style of field catalogue (Fig. 1) superior to the blank page because it prompts the wandering mind and results in more consistent quality and completeness of data. It can easily be generated on 100% cotton paper using a computer and laser printer. Using a loose-leaf catalogue format on standard-sized paper enables several individuals to use the catalogue simultaneously, makes photocopying easy (to distribute to project participants, for example), and allows completed originals to be immediately archived in the museum to prevent loss. The loose, unused sheets are kept together in a folding aluminum clipboard/container, and usually just one sheet at a time is removed for use. Com-

Kevin S. Winker Field Catalogue

KSW No.

Date: 10 June 1998 2330 Locality: British Columbia: Queen Charlotte Islands, Graham Island 53°23.7'N 132°19.4'W Mass: 23.1 g

Species: Catharus guttatus HAB: as 2326

WCH: 89.2 TL: 67.5 TS: 26.2 BL: 8.3 BLH: 3.2 BLW: 3.3 SKL: 34.6

FAT: v. lt. (TE)/OV: l = 9.4 × 6.3 SK: 100% ossified

MOLT: none STOM: insects

Remarks:

Age & Sex: AHY ♂ Disposition: skin, partial skeleton, stom, tissues (2) Collector/Preparator: K. Winker / "

Date: 11 June 1998 2331 Locality: " " Mass: 32.8 g

Species: Catharus ustulatus HAB: Second growth spruce-hemlock of 15-20 ft.

WCH: 98.5 TL: 72.2 TS: 28.9 BL: 9.5 BLH: 3.8 BLW: 4.6 SKL: 40.8

FAT: none (TE)/OV: l = 9.9 × 6.2 SK: 100% ossified

MOLT: none STOM: insects & seeds

Remarks: Came in to tape on blind playback. Topotype of C. u. phillipsi.

Age & Sex: AHY ♂ Disposition: " Collector/Preparator: " / "

Date: " 2332 Locality: " " Mass: 30.1 g

Species: " HAB: "

WCH: 99.7 TL: 72.3 TS: 29.1 BL: 9.4 BLH: 4.0 BLW: 4.4 SKL: 39.7

FAT: none (TE)/OV: l = 9.6 × 4.8 SK: damaged; see prepared skel.

MOLT: none STOM: seeds & insects

Remarks: " . " .

Age & Sex: AHY ♂ Disposition: " Collector/Preparator: " / "

Date: " 2333 Locality: " " Mass: 158 g

Species: Cyanocitta stelleri HAB: "

WCH: molting TL: molting TS: 43.1 BL: 22.7 BLH: 10.1 BLW: 10.9 SKL: 66.6

FAT: none (TE)/OV: l = 4.4 × 2.2 SK: 100% ossified

MOLT: Symmetric wing & tail, heavy body. STOM: insects

Remarks: Calling at passing raven, otherwise silent.

Age & Sex: AHY ♂ Disposition: " Collector/Preparator: " / "

FIGURE 1. A form-style field catalogue page improves the consistency and quality of specimen data. A loose-leaf catalogue on 100% cotton paper is easily made up and printed using a word processor and laser printer. It is also easy to make copies when the sheets are completed, and one can take only blank sheets into the field, offering less risk of losing completed pages and their data. Key to abbreviations used in catalogue and on labels (Fig. 2): HAB: habitat; WCH: wing chord (Baldwin et al. 1930:76); TL: tail length (Baldwin et al. 1930:92); TS: tarsometatarsus (''tarsus'') length (Baldwin et al. 1930: 107); BL: bill length from anterior edge of nostrils to tip (Baldwin et al. 1930:16; see Parkes 1988 for discussion on bill measurements); BLH: bill height from anterior edge of nostrils (Baldwin et al. 1930:20); BLW: bill width from anterior edge of nostrils (Allen 1889:188); SKL: skull length from rearmost part of skull to bill tip; TE/OV: testes/ovary; SK: stage of ossification (most useful among passerines), stated in approximate percentages.

pleted sheets and those in the process of being completed during specimen preparation are stored in filing cabinets in the museum and can be bound if desired.

Assignment of unique numbers (i.e., using a field catalogue) becomes necessary as soon as subsamples are taken or preparation begins. The unique number assigned to a specimen is a combination of the catalogue abbreviation (full initials if personal, a project or laboratory abbreviation if not), and an incremental number (Fig. 1). The field catalogue should contain all of the data associated with the specimen, and recording the appropriate data for each specimen should begin with collection and be steadily completed during the preparation process. Until preparation is complete, each entry in the field catalogue is a working document. The collector and preparator (often the same person) work in tandem (perhaps months or even years apart) to complete the entry.

The final label should be written immediately after preparation and the field catalogue entry are completed. The data associated with a specimen are what make it valuable to science. Completing the field catalogue entry and the skin label during and immediately following preparation (and before doing anything else) not only provides an immediate review, assuring that all of the appropriate data are recorded, but generates a second original record of these data, a prudent safety precaution. When finished, then, two complete original documents exist containing the full data for the specimen and its preserved parts.

To streamline data recording and information retrieval, I use two museum labels during the collection and preparation process. I consider it essential that full field data are attached to the specimen right after collection or salvage, and use the first label in the field, writing in (at least) date, locality, and collector. This label is later used for the partial skeleton following preparation; the only additions to it are sex, a note of the disposition of the specimen (how it was prepared), and a second writing of the field catalogue number in case the label becomes damaged in the skeleton preparation process (it may be partially eaten by dermestids if it is exposed to body fluids). The second label (a new one) is made up following preparation as the final label for the skin, and contains all of the data associated with the specimen. This final label for the skin is written as the last step in preparation and is tied to or pinned unambiguously beside the drying specimen. Always record a specimen's complete data before going on to another. Never try to remember these details to write them later.

At a minimum, a label should contain the date and locality of collection (or salvage) and the name of the collector. In montane regions elevation should be included. A temporary or field label with this information is all that is needed if a specimen is being frozen for later preparation, but additional information is often included at this time (e.g., soft part colors, behavior, habitat description). Such a label is far too incomplete for a final specimen, however. A complete specimen label should contain data on date, locality, collector, preparator, field number, sex (including con-

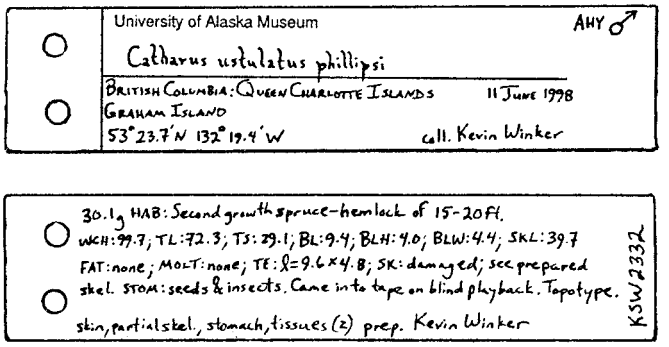

FIGURE 2. A completed specimen label. Actual dimensions are 19 × 86 mm. Note that it is flipped from front to back on the short axis. Abbreviations given in Fig. 1.

dition of gonads and any external reproductive characters such as cloacal protuberance or incubation patch), skull ossification (in species like passerines where it is a useful aging character; see below under Aging by Skull Ossification), mass, fat, molt, and disposition (what parts of the animal were preserved). Additional data are very desirable, but may be dispensed with if necessary: habitat where the bird was obtained, soft part colors (e.g., bill horn, iris red, feet lead blue, etc.), measurements, age, stomach contents, behavior, time of day, parasites, any noteworthy remarks, and species identification. The identification of a specimen is perhaps the least important, except for very young birds and complete skeleton preparations when no skin is preserved, in which case it is essential. Errors are regularly made in field and laboratory identifications; those preparing complete skeletons should preserve skin material as well for vouchers. Skins are identifiable to species and usually subspecies in the museum collection, and serve an important role as vouchers, especially for molecular studies (Winker et al. 1996).

There are standard ways to record most of these data so they are instantly recognizable and fully and unambiguously recoverable from every label. Figure 2 shows a widespread method. The positions on the label where the data are recorded are important also; following these standards assures that your labels will match those in most museum collections.

Date is given as day (number), month (written out in letters), and year (four-digit number). *Never* represent the month as a number! This error has caused many specimens to be removed from studies because of uncertain dates. Locality is written in full, with the major geographic division first, followed by greater detail and ending with the most specific information (preferably latitude and longitude). Collector and preparator should be clearly indicated (front of the label for the former, back for the latter) so the quality and veracity of the data can be judged and credit given to the people responsible for the specimen's collection and preservation. Field number connects all of the parts of the animal to the same

individual and points to the field catalogue entry where original data are recorded. The sex of the bird can only be truly learned by dissection, making the preparator fully responsible for accurately determining and describing the sex organ(s). This information is crucial, and is often accompanied by other sexually-related data (e.g., incubation patch or cloacal protuberance present). Body mass must be taken as shortly after collection as possible and prior to removing anything from the bird. A bird's mass is related to size, fat load, and reproductive condition, and is a useful datum for many types of studies. Fat load gives important clues to a bird's physiological or migratory condition, and molt provides additional important data about the condition of the individual and the life cycle of the population or species. If no evidence of fat or molt is found, write ''none.'' An absence of information means that you didn't look. Degree of skull ossification is useful for aging many passerines. Remarks provide additional information about the specimen; it may be the parent or offspring of another individual (give catalogue number), heavy parasite load, disease, or aberrancy; or it may have been washed in preparation. Finally, disposition informs the future user and museum cataloguer what else exists from this individual, greatly aiding recovery and use of all available preserved parts.

These constitute the essential data that should accompany each specimen, and it can be seen from Figure 2 that both the front and back of the label record this information in traditional positions. The rest of the data that most specimens should contain (e.g., habitat, measurements, remarks, etc.) are written on the back in the remaining space. I find that a free-form text method enables me to fit everything on a single label, even if six or seven lines are required. Using a stereotypical order duplicated in the catalogue (Fig. 1) speeds writing and information recovery. Our data entry software is also set up in this order to speed the computerization and cataloguing of specimens. A generic, preprinted label format (Fig. 2, without the writing) is preferable because it allows maximum flexibility and can be ordered in volume. Custom stamps are often used to fill in parts of the label when working for a long period in a particular locality; preprinted labels can also serve this purpose. Foster and Cannell (1990) discussed most categories of label data at greater length. Using a second label may be necessary occasionally, especially when descriptions of soft part colors are long, or when sketches of gonads, bills, or skulls are used. In these cases the back of the second label serves as another page.

The final label attached to the skin is an original document written by the collector/preparator to the future users of the specimen. It should be complete and it should retain its originality. Never replace an original label. If more space is needed for writing, add a second label. It is amazing how much can be learned of past preparators and collectors—their strengths and weaknesses—through reading their labels. Further, the samples of handwriting borne by each label are often essential for interpret-

ing comments subsequently written on labels (in pencil) in museum collections.

The labelling practice of affixing only a number to specimens, to have labels completed later (e.g., upon return from the field, or after preparation), is to be assiduously avoided. Having spent inordinate amounts of time trying to track down the field catalogues associated with such specimens, I can state assuredly that the small amount of time saved by the collector or preparator regularly dooms the product. An appropriately labelled specimen is immediately useful, regardless of its final destination and uncertain preparation, deposition, and cataloguing future.

A third label is eventually made up for the finally prepared skeletal specimen in the museum, but these labels are often computer generated and added to the specimen months or even years after the skin has been catalogued.

Labelling tissue vials presents a special problem in that, for frozen samples, labelling must be completed prior to freezing: one cannot write on a frozen vial. Some curators have gone so far as to give tissue samples final tissue catalogue numbers during preparation (in field or laboratory) by issuing blocks of blank tissue catalogue pages or numbers to collectors and preparators. This requires the preparator to fill out two catalogue entries during preparation, just to issue the specimen a (usually) second unique number. I find this to be needlessly cumbersome and rarely comprehensive—not all specimens entering a collection will be generated using the in-house numbering system. Cryovial manufacturers make inserts for the vial lids that can be written upon and inserted when samples are finally arranged for cryostorage and catalogued. A specimen does not need multiple unique numbers prior to being catalogued into a collection, and the traditional field catalogue number is sufficiently unique to serve the purpose. This number is also borne by all of the other parts of the animal. Although I haven't seen a duplication in the traditional initials-and-number field catalogue system (Fig. 1), the date, taxon, and locality information accompanying the number make later confusion seem impossible should duplication ever occur.

However, given the labelling constraints on tissue vials (even tissues in buffer or alcohol should be frozen for long term archiving), the method outlined in Figure 3 is preferred, since it provides all of the information necessary to assure rapid and accurate sample recovery from the freezer whether the sample has been catalogued into a collection or not. Writing the field number twice is insurance against the loss of information through abrasion, which is not uncommon, especially in LN_2 dewars.

Finally, no matter how rushed you seem to be, make a conscious effort when writing labels to do a slow and careful job. You are not writing notes to yourself. It is imperative that your penmanship be exemplary. For example, never write a label in script handwriting; always print carefully. Localities, remarks, strange abbreviations, and people's names are often uninterpretable on specimen labels because someone was in a hurry.

Temporary fluid preservation.—Freezing specimens is not always possi-

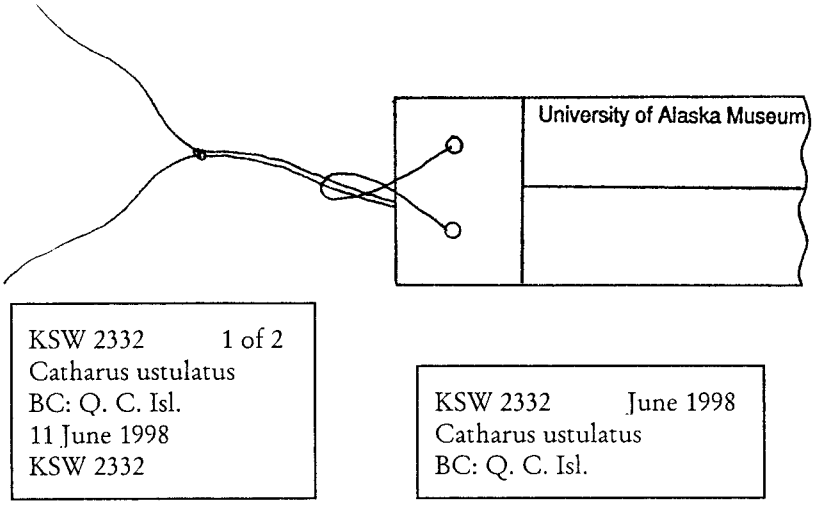

FIGURE 3. Label as written on outside of cryovial of tissue samples, stomach contents vial label, and traditional skin label knot.

ble. Yet, when one has only a short time in the field, preparing every bird as it is obtained would seriously limit the number of specimens that could be collected. This is particularly true for brief visits to remote locations. Weber et al. (1984) presented a method of fluid preservation that enables one to maximize field time in such situations and bring specimens back to the laboratory for preparation. Specimens preserved this way are more difficult and time consuming to prepare, and on average the skins are not as good (aesthetically) as those freshly prepared. However, the method enables one to collect more individuals when in the field, increasing the return on the investment. I have had some success with this method of temporary preservation. It involves a dilute solution of formaldehyde, phenoxyethanol, and salts. Concentrated chemicals are carried into the field and mixed with water in a pickling container. Gloves are used whenever contact with the preservative is possible, and care should be taken to minimize inhaling the low-level formaldehyde fumes.

Tissues are removed from the specimen prior to pickling (formaldehyde causes serious damage to DNA; Vachot and Monnerot 1996) and are placed in buffer or LN_2. The bird is sexed, eviscerated, and labelled, its stomach preserved in alcohol, the plumage and body cavity cleaned of blood, then it is wrapped and pinned in a single layer of loose gauze prior to immersion in the pickling solution. Do not use pins with plastic heads; the heads dissolve and may stain the plumage. When the specimen is pickled its position should be close to that of the final preparation, because the feathers become essentially pickled into place and are far less workable than in a fresh or frozen specimen. Blood should be assiduously removed prior to wrapping in gauze; it becomes fixed in the solution and

stains the plumage with a dark, blackish-brown color. Washing bloody plumage in clean water and swabbing out the body cavity with cotton prior to wrapping are recommended.

Specimens should be stirred once or twice daily in the solution to assure complete pickling. Upon leaving the field, the fluid can be drained and the moist specimens wrapped in cloth and placed within many plastic bags and/or a leak-proof container for transportation back to the laboratory. There, a fresh solution can be made up to complete any pickling not finished in the field, as well as for storage of the specimens until preparation. I have detected no change in such specimens stored this way for up to 4 yr following collection. Prior to preparation, the pickled specimens are rinsed under clean water for 1–3 d to leach out most of the chemicals. Gloves are used in skinning, and each skin must be completely washed and dried.

Soft part colors.—The colors of the bill, legs and feet, irides, and things like wattles, external air sacs, etc. generally change rapidly upon death. The colors of these parts often vary among age and sex classes as well as seasonally and geographically. Describing these colors while the bird is still fresh using standard color references such as Ridgway (1912), Smithe (1974–1981), and Munsell (1990) enhances the value of the specimen both to science and to artists who might use the specimen for subsequent illustrations.

Measurements.—Many specimen measurements change upon preparation and drying (e.g., Winker 1993). A bird should be weighed and measured before anything is modified or removed (e.g., tissues or stomach). Occasionally, one encounters birds whose mass is clearly affected by substantial food items; such individuals can be re-weighed with the food items removed (note this in Remarks). A researcher may take numerous measurements from a prepared specimen, and it is not up to the preparator to anticipate these. However, there are several commonly taken measurements that change or are unobtainable in the completed specimen. I generally take seven measurements prior to preparation (Fig. 1). Taking these properly requires practice, but they can be performed rapidly and accurately, with precision to 0.1 mm when using a vernier caliper is possible. Further discussion can be found in Winker (1999). Units of measure are always grams for mass and millimetres for morphological characters, and I omit writing ''mm'' for morphological measurements.

TOOLS OF THE TRADE

Although a perfectly good specimen can be prepared using nothing more than a good pocket knife, some cotton balls, and makeshift materials, proper tools speed the job considerably. A sharp scalpel, two or three sizes of forceps, small and large scissors, vernier calipers, a millimetre tape measure (for larger birds), pens with black, waterproof ink, cotton paper and labels, heavy and light thread, needles, and pins constitute a good preparation kit. In addition, cryogenic vials, vials for stomach contents, pinning boards (e.g., styrofoam), gauze, cotton, sticks, cob dust (or saw-

dust or cornmeal), excelsior (for large birds), heavy thread and string, and alcohol constitute supplies consumed in the preparation process. Some sources for supplies are given in the Appendix. Finally, a personal, project, or laboratory field catalogue should be available for recording full data and assigning a unique catalogue number to the specimen.

No single pen functions well for all of the writing that preparation requires. *Never use pencil.* It doesn't xerox well (important for field catalogues), it often fades or smudges, and it can completely disappear with grease and long term label color changes. Only black, waterproof inks should be used, and your pens should be tested for known short-term problems. For example, alcohol may dissolve a waterproof ink, which is important to know before putting labels in stomach content vials. Technical pens, used by draftsmen and artists, are ideal for labels and field catalogues, but are expensive and inferior for cryovial labeling because the ink flakes off at ultracold temperatures. Fine point permanent markers (e.g., ''Sharpie'') are good for writing on cryovials, but are usually inferior for labels and catalogues, and they usually dissolve with alcohol. Inexpensive ballpoint pens with permanent ink work well for field catalogues, but can rarely be found with point sizes small enough for the delicate writing of legible labels. And some ballpoint inks dissolve on fat-soaked labels or when exposed to naphthalene (K. C. Parkes, pers. comm.). I use three pens: technical for labels, permanent ballpoint for catalogues, and permanent marker for cryovials.

PREPARATION

1. Put a wad of absorbent cotton or facial tissue down the throat to prevent fluid leakage. If one is already there, replace it.

2. Record soft part colors if bird just died (Ridgway 1912, Smithe 1974–1981, Munsell 1990).

3. Weigh bird and take any desired, irreproducible measurements if not already done. Record in catalogue. Note if bird has been in freezer for a long time prior to weighing; desiccation causes loss of mass.

4. Label tissue cryovials with Sharpie marker. Never use a technical pen. Include catalogue number, species, locality, date, and catalogue number again (Fig. 3). Abbreviate species and locality if necessary; make sure the catalogue number is written legibly and indelibly *twice*; they can become lost through abrasion or illegibility. Usually, take two tissue samples. Label vials as ''1 of 2'' and ''2 of 2.''

5. Make incision in skin from furculum to cloaca. Begin separating skin from body (Fig. 4). After making the initial incision, separate skin from body using forceps to push body surface inward while gently pulling skin outward or, better, pushing skin outward using forceps and/or a finger (Fig. 4).

6. Make incision into body cavity; carefully remove stomach/gizzard (big, hard, in the way) by tearing or cutting the esophagus and intestine free (Fig. 4). Set it aside for later.

7. Be careful not to disturb or mess up the body cavity too much: you

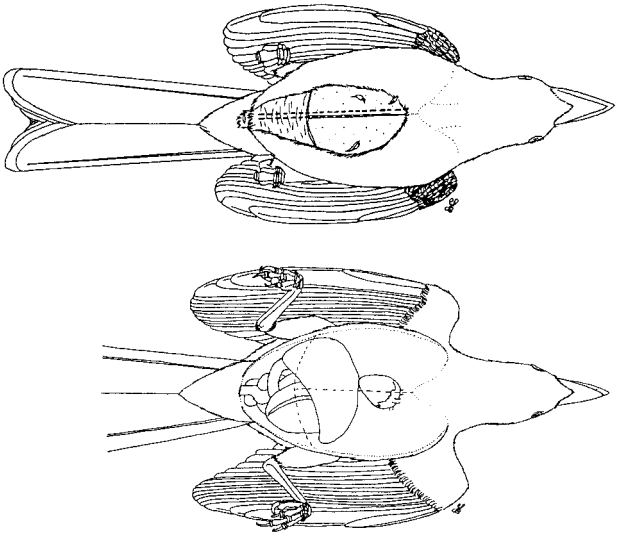

FIGURE 4. Initial incisions through skin and abdomen and relative positions of major organs occupying the abdominal cavity, illustrated, anterior to posterior, as heart, liver, stomach/gizzard, and intestines.

must be able to examine the sex organs, which lie along the dorsal surface of the cavity near/upon the anterior side of the kidneys (Fig. 5). Intestines can usually be removed—carefully, so that fluids don't get on feathers and gonads aren't disrupted.

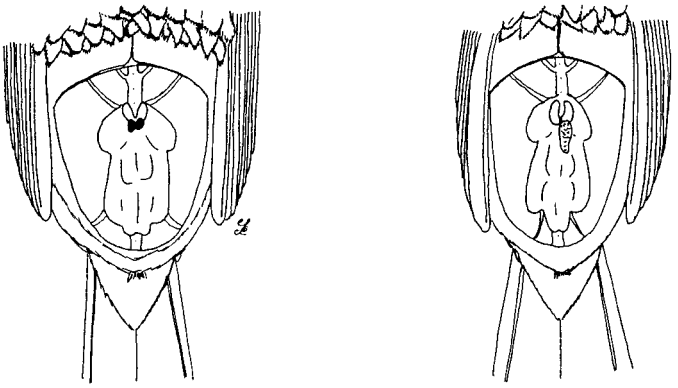

FIGURE 5. Determining sex requires direct observation of gonads, here illustrated in a nonbreeding male (left) and female (right). Gonadal tissue almost always lies on top of the posterior third of the bird's left adrenal. Adrenals occur in both sexes in pairs near the anterior edge of the kidneys. Males always have two testes, but most females have only one ovary, the left, as illustrated.

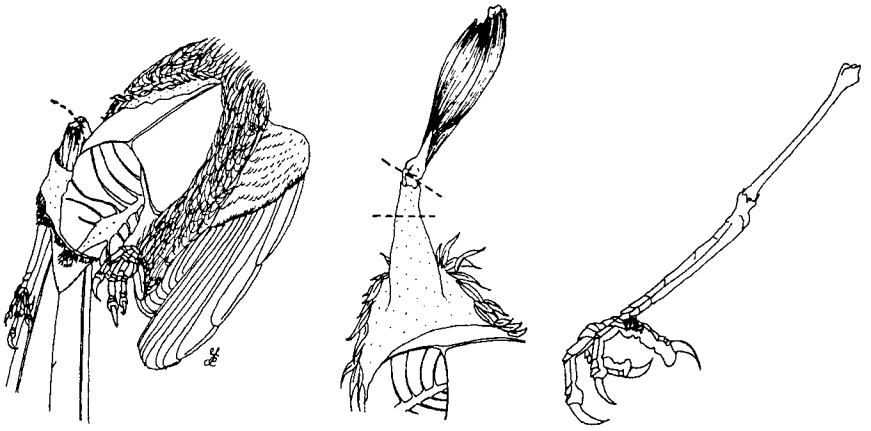

FIGURE 6. Poke the knee into visibility and disarticulate. Skin leg to next joint (articulation of tibiotarsus and tarsometatarsus). One leg should be removed for the skeleton by cutting the skin (center, lower dotted line; cleaned leg at right). In some cases disarticulation of a leg is required to obtain one unbroken tibiotarsus for the skeleton (center, upper dotted line).

8. Take tissue samples and sex bird. First remove heart (Fig. 4). If from a small bird, place whole heart in cryovial 1. If bird is larger (e.g., *Catharus*, *Passer* or larger), cut heart in half and put half (or some) in each vial. Next, put some liver into each vial, then sex the bird (see Determining Sex below; Fig. 5). Record gonad type and size in the catalogue. After sexing, kidneys can be removed (this disturbs the gonads); put some kidney tissue into each tissue vial. At this point your tissue vials should each be approximately ½ full, except for very small birds (e.g., many Parulidae, Sylviidae, Trochilidae). Fill each to within ca. 2 mm of top with breast muscle. On small birds this involves fileting out the entire breast with a scalpel. Replace caps onto vials very tightly and freeze tissues immediately. Caps that are not replaced very tightly often come off in LN_2 and the samples are lost.

When you have attained the ability to skin out a bird in about 10 min (separating skin from body), you can take tissues after the body is out. Taking tissue samples and sexing a bird is much easier with the body removed, but tissue freshness is extremely important. I try to get tissues frozen within 30 min or less of thawing to maximize their usefulness in molecular studies. With the carcass skinned, more tissue can be taken in small birds. When skinning small birds, I generally take all large muscles (breast, wings, legs) and all internal organs except intestines and stomach. Lungs and kidneys must be lifted up out of their bony recesses, and with practice can usually be taken out in one or two pieces.

9. Skin. Begin with knees, and proceed with legs (Fig. 6), tail, and shoulders, to head (making incision in rear of head/neck for large-headed birds such as Picidae and Anatidae), then pinch and roll out ears, and

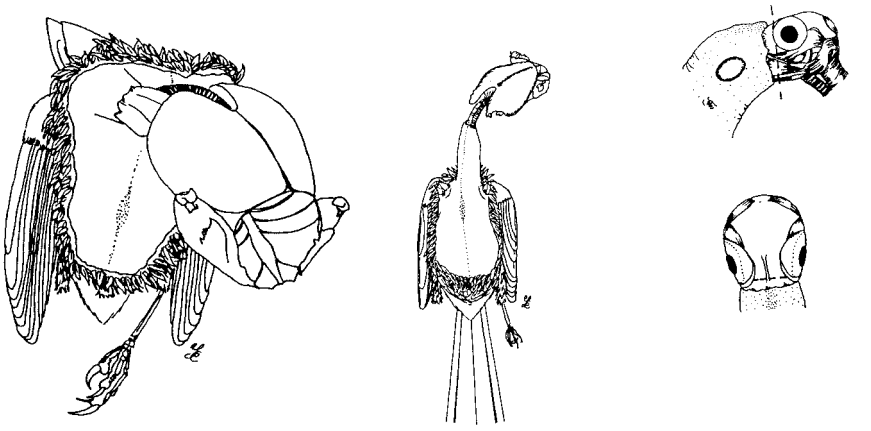

FIGURE 7. Body is skinned to shoulders, which are disarticulated. Skinning proceeds to head, then past the ears, which are pulled free, and then past the eyes, from which the skin is carefully cut behind the eyerings, to the base of the bill, where scissors are used to cut the skull free in a plane across the anterior portion of the eye sockets (dotted lines). Care must be used not to cut the tongue, which should come free with the body for the skeleton.

cut skin away from eyes back far enough so that eyering remains intact. Skin can be separated from body to base of bill, just in front of eyes (Fig. 7). To minimize holes, use forceps and fingers to separate skin from body. Scalpels are used only where it says ''cut'' in details below. I use scissors only to cut the vertebrae at the tail, to make the skull cuts, and, later, for cutting thread.

Details: After the initial skin incision (step 5), work on one side at a time. In general, throughout the skinning process you wish to avoid pulling on the skin. Instead, work at the point where the skin still adheres to the body, pushing the skin free using a thumbnail or forceps where possible. Poke the knee (by holding the lower leg) in toward the body while holding the skin away from the body (sort of lifting the abdomen skin in a bunch over the knee while the latter is held and pushed inward). Pop the knee into visibility (Fig. 6). You don't need to see much of it. Using a wiggling motion (rather than slicing), ''worry apart'' the knee joint and the surrounding muscle.

After the leg is detached below the knee from the body (attached only to leg skin), push the skin free with your thumbnail down to the ''ankle'' joint (Fig. 6). This is the joint between the tibiotarsus and the tarsometatarsus, and in most birds it is where feathering ends and skin or scales begin. One leg will be removed and saved for the skeleton—the right if unbroken. To remove one leg, when reaching the ''ankle'' joint cut a ring around the skin to separate it from the limb and remove the whole leg for the skeleton (Fig. 6). If the foot is too large, cut the muscles away from the tibiotarsus to get it out through the hole you've made at the

base of the feathered leg skin, or make a lengthwise incision in the leg skin to enlarge the hole at its end. Remove the muscles from the tibiotarsus and, if the bird is very small, add them to a tissue vial. Set this removed leg aside for the skeleton (Fig. 6), and put the leg skin back right side out. On the remaining leg, cut through the knee joint and skin down to the ''ankle'' joint again. Now remove the muscles from this tibiotarsus, wrap a little cotton around the bone to replace the muscle mass, and gently pull the leg back inside right and quickly brush the feathers back into place. Leaving this bone in the skin with its cotton wrapping adds a lot of strength to the leg and makes it comparable to historic skins. If you have already saved a whole leg for the skeleton, cleaning the muscle from the tibiotarsus remaining with the skin can be performed very rapidly by breaking this tibiotarsus near the proximal end, peeling the whole muscle mass away from the bone, cutting the tendons in one slice near the distal end, and using a little dust and two fingers to rub away any small amounts of meat remaining.

If a leg (tibiotarsus or tarsometatarsus) is broken, it stays with the skin. If neither leg is broken, the left will remain with the skin. Decisions about which bones stay with the skin and which go with the skeleton are made before cutting the knees and after carefully feeling and visually examining each leg. In most cases the left leg can remain with the skin. Be sure that one whole bone of both tarsometatarsus and tibiotarsus is available for the skeleton. In some cases (commonly in shot birds) it is necessary to disarticulate the tibiotarsus from the tarsometatarsus that will remain with the skin to obtain a complete tibiotarsus for the skeleton (Fig. 6). In such cases the remaining leg is only attached by a little bit of skin. It can be given extra support through replacing the missing tibiotarsus with the addition of a small stick and some cotton, but I generally skip this in small birds to save time.

Now, working on each side separately to prevent the skin from being stretched and ripped, separate skin from around the pelvic area. Work down into this area; don't try to open it up and lay the skin out flat or you'll tear it. When the skin is relatively loose down the sides, hold the tail base between thumb and forefinger, putting the forefinger forward enough to cover the muscular tail base and serve as a stopping point for your scissors as you cut the vertebrae from the inside. From above, cut the lower abdomen posterior to the upward-projecting public bones and aim your scissors to cut the vertebrae behind these bones and in front of the muscular base of the tail. Care must be taken not to cut the bases of the tail feathers or they will fall out; however, the synsacrum or pelvic bones should not be damaged either. When the vertebrae are cut, use a scalpel to free the remaining tissue so the tail is separated from the body. Free the lower body from the skin and evert skin with fingers to the shoulders, disarticulating these with the scalpel (add dust). When skinning down the back, push skin away from body using thumbnail and take care not to tear the skin by stretching it too much around the body. It often helps to get it off of the high point of the breast as soon as possible.

This can easily be done by grasping the whole skin-body interface all around the bird with all five fingers simultaneously and pushing-pulling this skin mass past the broadest body area. Disarticulate the shoulders to separate the wings from the body (Fig. 7). This loosens things up considerably. Be sure to add a lot of dust during this phase or you'll get fluids on the feathers.

Continue peeling to head, making an incision in rear of head/neck for large-headed birds such as Picidae and Anatidae (see Variations below). With a little dust for friction, pinch and roll out ears, which prevents any holes being made, then cut skin away from eyes back far enough so the eyering remains intact. Skin can be separated to base of bill (just in front of eyes). Using scissors, cut skull away from skin at the base of the bill. Use three cuts, and don't cut the tongue: a) Upper, frontal portion of head, done straight down from the top of the head with the scissors in the frontmost area of the eye sockets (don't cut through the eyes themselves); b) Left mandibles: insert scissors (sharp tip inwards helps) straight upwards on each lateral side of the upper and lower mandibles to cut these bones; c) Right mandibles, same as left (Fig. 7). Gently twist and pull to finish separation, keeping in mind that the tongue comes with the body. Also, don't pull very hard, or you'll rip the bill away from the skin. You can make auxiliary cuts if necessary through the palate, but keep the tongue (and its bones) intact. The body should now be separated from the skin. If you haven't taken tissues and sexed the bird, do so now (see Determining Sex below; Figs. 4 and 5).

General points: Always be sprinkling corncob dust, sawdust, or cornmeal onto any fresh surface to keep fluids and stickiness from getting onto feathers. Feathers must be kept from contacting body fluids or you will have to wash and dry the bird—a time consuming task to be avoided whenever possible. Whole bones are removed for the skeleton until one of each is preserved; broken bones can remain with the skin.

10. Skin, clean, and tie wings (Fig. 8). Setting the body aside, sexed and with tissues taken, take up the skin and skin out the wings (Fig. 8). Begin by getting the shoulder muscles up away from the skin so you can peel down the humerus, then work the skin away down to the joint where the radius and ulna begin. The secondaries are attached to the ulnae, and they must be separated carefully to prevent tearing. When skinning down the radius and ulna, place a thumbnail directly onto the ulna and forcibly push downward (or outward, toward the distal end of the wing) to separate each secondary from its bony attachment. In very large birds a dull knife may be necessary to help make this separation. Skin down to the ''wrist'' joint (where the radius and ulna form a joint with the ''hand'' bones), and cut through this joint, having skinned down far enough that you don't cut the skin. Skin the other wing similarly. Leaving one wrist intact is desirable so that the skin is comparable in wing measurement to the great number of historic specimens. Make the choice based on skeleton quality: if the radius and ulna are broken on one side, leave the distal portions of these bones with the skin. If the bones on both sides

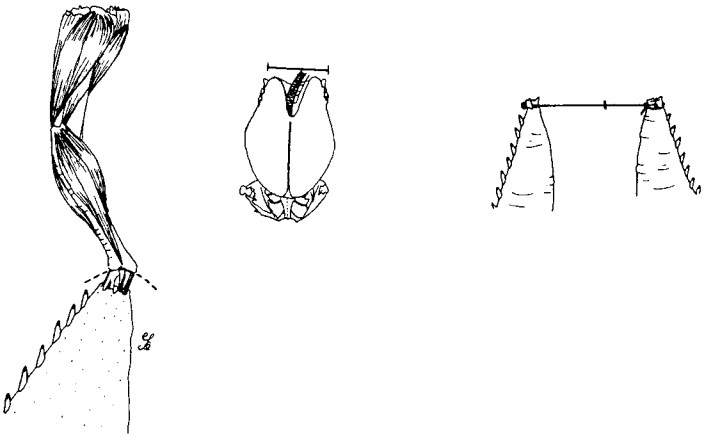

FIGURE 8. Skin wing to distal ends of radius and ulna, separating bases of secondaries from ulna with thumbnail. With wings cleaned and bones removed for skeleton, consider interscapular distance on the carcass as one unit (center) and firmly tie wings together so that the wrist stubs are 1 ⅔ units apart.

are unbroken, I leave the left wrist intact, breaking the left radius and ulna in half (a traditional method) and leaving the distal half with the wing. In large birds, skin out the hand bones of one wing; I do the right. This must be done carefully, because the primaries are attached to these bones; the most distal bone is the most difficult. There is very little skin down here, so work carefully. With practice, all of this becomes easy.

Thread a rather heavy needle with heavy thread (I prefer white button and carpet thread, used also for labels) and make a good, big knot at the end of the thread. With the distal ends of the wings still everted, push the needle through the ''hand'' bones (but not the skin) of one wing and pull the thread through to the knot so it's tight. The other wing can be tied with the thread to the broken ulna, or push the needle through the other wrist, making sure first that the two wings are oriented correctly (with their dorsal surfaces facing inward; Fig. 8). If the hand bones are too sturdy to be run through with a needle, the wings can be tied together by running the needle through skin on the underside of the wing beside the distal joint of the radius and ulna. Be careful to use the underside, where disarrayed feathers are hidden in the final skin, and not to put the thread too close to the bases of primaries or secondaries, which often become twisted as a consequence.

Now, arrange the everted wings so the dorsal surfaces face each other and the primaries extend straight backward, almost touching each other. Hold the two everted wing stubs in one hand with your first three fingers at a distance from each other equal to the distance between the scapulars on the carcass (we will use the carcass for a model several times). This is ''one unit''—the distance between the two wings on the living bird (Fig.

8). Pull the thread so it is taut between the wing stubs when they are held at this distance (it is held on one side by the knot, on the other side by your other hand). Now, with a pair of forceps, pull back into the inter-stub space enough thread to equal ⅔ of the ''one unit'' of distance you continue to maintain between the two everted wing stubs. Carefully tie off the loose end of thread so that the two everted wing stubs are firmly tied together at 1⅔ times the ''one unit'' distance (Fig. 8). If tying to the broken ulna on one wing, use about 1⅓ interscapular ''units.'' This distance is important: if you're off, the skin will be difficult to arrange properly later.

11. Clean tail, being careful of the thin skin. Be sure to cut or scrape out the uropygial (oil) gland on the dorsal surface. Also, don't cut off too much muscle or other material here, especially in small birds, or tail feathers will begin to fall out. But you do want it relatively clean and grease-free. When most of the meat and glands have been scraped away, use dust to rub remaining meat and oils off.

Between skinning and cleaning up the tail, a lot of work is done in this area. In small birds this is a difficult part of the job—you are working in a tight space where there is little skin and little room for error. You don't want to tear off the tail (easily done in small birds), or to have tail feathers fall out (also easily caused). Conversely, you have to get out the uropygial gland and most of the meat so the preserved skin isn't ruined by rot or oil leakage.

Tail feathers are arranged at their bases in tight rows whose basal tips outline a rearward-projecting arrow. They are attached to the pygostyle, a long, narrow bone that projects posteriorly down the middle of the two rows of tail feathers. This bone and most of the muscle (all of the muscle in large birds) can be removed (if done carefully) without compromising the integrity of the tail feathers and the skin surrounding them. In small birds, I generally evert the base of the tail, then hold it by folding the tail back against the outside, dorsal surface of the skin, then grasping the tail and the skin firmly together in one hand. This prevents stress on the little skin keeping the tail attached, and the tail stub is exposed for careful work with a scalpel. Meat edges are carefully teased off of the skin with the scalpel edge and the skin worked back off of the tail stub on all sides using a thumbnail. On the dorsal surface you need to expose most of the uropygial gland, a white or yellowish bilobed organ. Usually you can't expose the whole organ without tearing the skin, so be careful. Cut and scrape all of this organ away (some scooping action is often necessary to get back under the skin). A few short feathers are present where this organ connects to the surface (this is where the bird wipes its bill to oil its feathers); cut this small ''nipple'' away from the inside or you'll make a hole. With the tail stub skinned, remove the remaining muscle by whittling it carefully away. Some of the remaining bone can be easily cut or broken away, but it is best to leave the pygostyle itself present in small birds—it keeps the tail feathers firmly in place and too often the tail is damaged in trying to remove it. Don't cut the bases of the tail feathers

(rectrices)! Give the cleaned tail and surrounding skin a dusting and rub to clean it further and dry the surface, then put things back inside right.

12. Fleshing. Scrape off any remaining meat or fat from the inside of the skin. This can be a very rapid step (e.g., in passerines with no fat), or a very long, drawn out process (e.g., in migrating waterfowl and seabirds). For fat birds, special tools such as toothed spoons, scraping wires, and wire wheels can be of great assistance in fleshing. With very greasy birds it is often best to wash them thoroughly after fleshing, first in soapy water, then in a solvent that dissolves fat (e.g., white gas, mineral spirits, paint thinner, hexane). Such cleaned skins dry up very nicely. Oils left in the skin leach out onto the plumage and also acidify over time, hastening deterioration of the skin. Well-fleshed skins and formerly fatty birds that have been washed in a fat-dissolving solvent are some of the best-preserved in museum collections.

At this point it is possible to put everything aside for awhile. Making sure all relevant data have been recorded, the skin can be moistened and frozen double-wrapped in a plastic bag *with a label* attached to the bird. I leave the head inverted (except for species with sharp bills or long head feathers), but put the rest feather-side-out (especially the wings), then place the bird with its label into a plastic bag, squeeze out all the air, and bag it again before freezing. Some workers place moistened paper towels or tissue against the skin in the cavity before freezing. The skeleton can be made up now or set aside as the skin was.

13. Put the skin back inside right. Before putting the skin right side out, moisten it thoroughly so that it is maximally flexible and each feather can be adjusted (not possible with a dry skin). Saliva and fingers, or a dish of water with a paintbrush or cotton swab can be used effectively to put moisture exactly where it is needed. Moisten the head well, but be careful not to get water on any feathers (especially through the eyes or other holes). Don't put on too much water—you just want the skin to be limber. Dust the skin thoroughly to take off the stickiness, then put it back inside right. The tail should already be reverted. Next revert each wing to feather side out, spreading out the skin and feathers on each side and rearranging the flight feathers and coverts into their proper positions. No bunched-up skin should remain anywhere between the wrists of each wing (to minimize wing feather disarray, revert wings before freezing if stopping before step 13). I should mention that it is fast and convenient to ''strip'' the secondaries from the ulna, but that for this convenience we must be assiduous in putting the secondaries back into their proper position. Beginners often fail to do so, leading some authorities (e.g., Van Tyne 1952) to condemn the practice. Now is a good time to check whether you're satisfied with the distance between the tied wings and to change it if necessary. To pull the head back through the neck, push the bill along until you can see and grab it from outside. Pull it carefully out and begin rearranging feathers generally.

This is the time to carefully inspect the plumage. Notes on molt and fat, much of it best seen while the skin was everted, should be made now.

Molting body feathers are easily seen on a fleshed skin as dark spots or short lines, which represent the blood associated with growing feathers. Flight feather molt must be assessed from outside. Details on fat are discussed below.

14. Washing. If dirt, blood, or other body fluids are present on the plumage, you will have to wash these places clean. Use a mild soap and water, and wash the dirty spots well. Rinse well, then dry. When only small areas or the head require washing, restrict the washing to these areas. Drying a bird is a time-consuming process, so spot-washing should be employed whenever possible. When the area (or bird) is well washed and rinsed, it should first be patted dry with some toweling, then thoroughly dried with forced air or with a combination of dust and forced air. In the lab, the forced air is usually compressed air or, for larger birds, an electric hair dryer (set on warm only—never hot). These tools speed the drying of washed birds considerably and make laboratory preparation preferable to field preparation whenever specimen washing is required. In the field, the forced air is from one's lungs.

When you have an adequate amount of dust, and when you are not working with downy birds (e.g., owls), you can dry a washed bird relatively quickly by shaking it about in a large, closed bag of cob dust. With a few double handfuls of cob dust in the bag, close it and firmly shake it about for 10–15 minutes, occasionally opening the bag and manipulating the skin to knock off sticking wet dust and allow dry dust to enter wet areas. In the later stages, remaining wet areas should be manually treated with dust, brushing dry handfuls repeatedly over the wet feathers. When it is nearly dry, remove the bird from the dust bag and shake most of the dust free. Then, in a well ventilated area where airborne dust won't offend people or equipment, blow the bird clean and dry using forced air or by blowing on it. If you don't do this carefully, you'll develop a ''dust cough'' from inhaling too much cob dust. A bird is dry when its downy underplumage is fully fluffy again. Note that it takes physical stimulation during the drying process to get the feathers to fluff up again. Much of the drying process consists of providing this stimulation by rubbing, thumping, or beating the drying feathers.

A ''dry wash'' is sometimes possible when mud or blood is present in small amounts on generally dark plumage. In these cases, the dirty plumage can be held firmly in one hand while with the other hand clean, dry cob dust (or cornmeal or sawdust) is rubbed firmly and repeatedly onto the plumage with the grain of the feathers. White or very pale plumage generally can't be cleaned this way. Dried blood or mud can often be chipped out by spot-rubbing with a thumbnail, with finer particles brushed away using a stiff brush (e.g., toothbrush).

15. Build and insert a three-piece cotton head: two rolled eyes the size of the bird's eyes (the whole eyeball, compared in the orbit with the cotton one; Fig. 9), and a piece of cotton formed to resemble the skull without eyes in the orbits (Fig. 9). Preen the feathers and shape the head while the forceps holding the last piece (the cotton skull) are still insert-

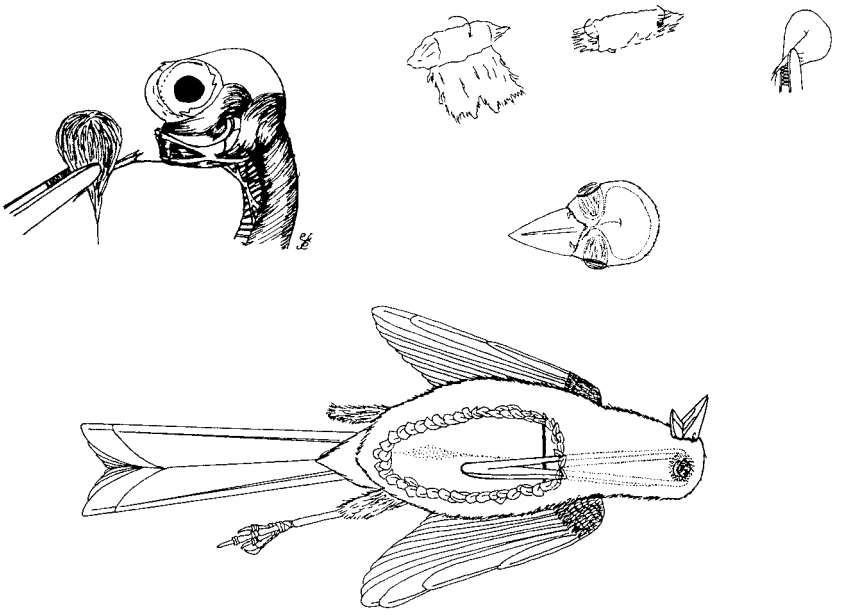

FIGURE 9. Shape smooth, firm, cotton eyeballs roughly the size of the original (compare with carcass), and firmly insert first one, then the other. Next, roll a cotton skull in a size and shape similar to the original skull without the eyes; firmly insert and arrange head skin and plumage with other hand before releasing cotton.

ed. It is also easiest to arrange all of the pieces separately, and as a whole each time one is placed inside, by working from inside with the forceps still holding the cotton and from the outside with your other hand. Specific and subspecific identifications frequently rely on characters around the eyes—make skins slightly bug-eyed. Symmetry is a strongly encouraged objective here.

16. Roll a cotton body onto a stick with a sharpened point, using the carcass as a model and making sure the neck isn't too long (Fig. 10). You want a sturdy stick that is at least the total length of the final specimen. Having a stick that projects to the tail tip protects against tail damage and provides a sturdy anchor for the leg and label. Wet the stick so the first cotton sticks to it, then wind long, narrow, thin pieces of cotton rather firmly onto the stick with one hand while you rotate it with the other. In this winding you are trying to mimic the length and girth of the main body of the bird, which you should have in front of you for comparison. Ignore the neck of the carcass, and make the body begin down from the pointed end of the stick approximately the distance from the nostrils to the base of the skull. Except in very long-necked birds such as waterfowl and shorebirds, in which a neck of cotton must be built, in the final skin you want the head cotton to rest against or very near the cotton body.

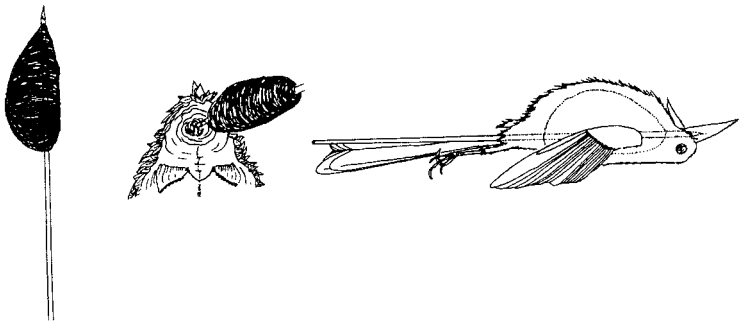

FIGURE 10. A cotton body is rolled in narrow, thin pieces onto a sharpened stick to resemble the size and form of the original body without its neck; the symmetrical rolled body is pressed into an asymmetrical shape resembling the original. The sharpened stick is inserted up the neck and into the remaining spongy bone of the upper mandible, and the skin is pulled onto the body.

Leave only enough stick emerging from the cotton body to allow this to occur. When the length, girth, and rough shape of the carcass are mimicked by the rolled cotton body and the surface is generally smooth, make one side the dorsal surface and the other the ventral by compressing one side and pushing cotton up onto the other, obtaining a final shape very similar to that of the carcass (Fig. 10). This enables the final skin to lie flat on its back, with the bulk of the cotton lying in the ventral portion of the skin, and prevents the skin from rolling in the museum tray.

17. Insert the pointed end of the stick into the skin (Fig. 10). Use forceps to help open the passage to the mouth (made easier by dusting away surface stickiness prior to reverting the skin). When you have the pointed end of the stick coming out of the mouth, withdraw it to the point where it goes over the edge of the severed base of the bill, then ram it into the bone of the upper mandible, aiming roughly for the stick to pass through the spongy upper mandible bone to come to rest in one of the nostrils. Get the bill on straight, and don't plug up the nostril or it will be difficult to get a bill measurement from the skin. Preen the head again.

18. Laying the skin and partially inserted body ventral side up on the table, use forceps to compress the cotton body locally and draw the skin up onto the body with your fingers, working carefully and evenly until the body lies in the skin, essentially in its final position (Fig. 10). Any adjustments to size must be made now. If the body is too large, take cotton off. If it is too small, add cotton in smooth, symmetrical contours where necessary. A small piece of cotton inserted into the skin pocket at the base of the tail firms it up. You should now be able to put the ventral incision edges back in their proper relative places with only slight compression of the cotton body.

19. Sew the skin back up. Tie leg to stick. Close bill. Bill must be both pinned and tied. The lower mandible must be put into its proper place

against the upper mandible, and is best held there with a pin through the base of the gonys into the upper mandible. This maintains horizontal alignment. A thread passed through the nostrils with a needle and tied around the bill and behind the pin (rather tightly, but not crushing light-billed birds) will keep it closed. In species with closed or inaccessible nares, or species in which the operculum is of taxonomic value (e.g., some swallows), an alternative method of bill closure is to push the pin completely through the bill and pass the tying thread around the bill behind the pin on both the dorsal and ventral surfaces. For long-billed birds such as hummingbirds and shorebirds, a second tying out near the tip (without a pin) assures permanent bill closure along its entire length.

Few stitches are required to close the ventral incision. Sewing should always be done by bringing the needle up from the inside of the skin through to the outside. I generally make about 4 loose, looping stitches in passerines, ending on or beside the cloaca. Closing the ventral incision is usually a rapid process that leaves gaps through which the cotton body is visible. Plumage arrangement covers this in most cases. The ventral incision should be carefully closed with closer stitches only in species with dense ventral plumage (e.g., waterfowl), in individuals with a bare venter, and in specimens that have been temporarily fluid preserved. When the ventral stitches have been placed, insert forceps below the stitches and on top of the cotton body, compress the body and, holding the needle in one hand and forceps in the other, draw the stitches firmly but not tightly closed in one pull. Holding the forceps in place, I next arrange the ventral feathers with the other hand to be sure everything looks fine, then hold and compress the whole body while carefully slipping the forceps out. The thread is then tied firmly to the stick. Before cutting it free, I make sure that the tail is centered and doesn't flop excessively when the specimen is lifted. At this point one or two additional stitches in the skin at the base of the tail can center it and hold it firmly against the stick if required.

The remaining leg should be tied to the stick midway along the tarsometatarsus with the leg in a relaxed, ''natural'' position that leaves it with the sole of the foot pointing toward the tip of the tail and with the tarsometatarsus fully exposed and available for measurement (e.g., passerine in Fig. 11).

20. Preen and rearrange feathers. Most feathers are best rearranged by working from their bases. Long, narrow forceps and in some cases a long rigid wire are very effective; brushes are not. In many species with narrow tracts of feathers on the neck (e.g., most passerines), the neck skin usually must be unstretched and slightly bunched (as in the natural bird) by pulling the skin near the bend of the wing upward towards the cheek; also try pulling a little skin downward from just below the cheek toward the bend in the wing. Tuck any visible plumage bases into their proper places below surrounding contour feathers and arrange these contour feathers over the bases still exposed. If the neck of the cotton body and stick was too long, you can try to push the whole body forward to un-

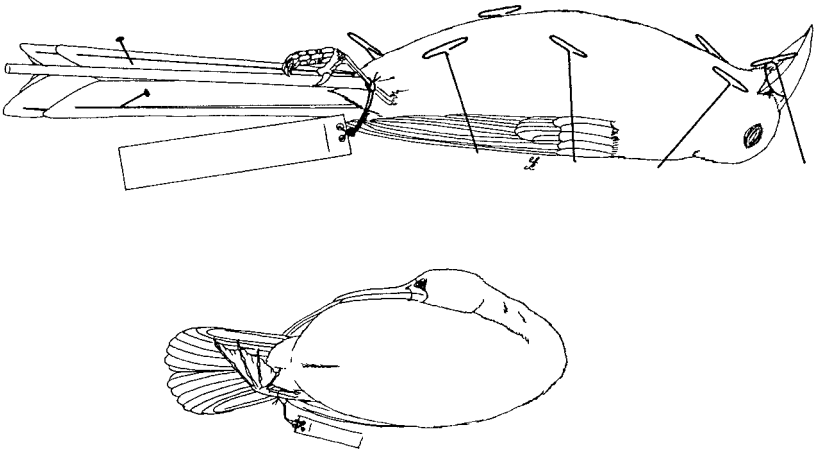

FIGURE 11. The sewn skin's plumage is carefully arranged, then the whole is pinned out in final form to dry. Long-necked birds should have their necks bent to shorten the specimen, minimizing space required for packing and in the museum tray.

stretch the neck skin; this is best noticed and done before the stitches in the ventral incision are tied off. Twisted feathers disrupt or break the smooth contours of the feather surface, and can usually be untwisted with forceps. Wings and the thread tying them together must sometimes be hiked forward into their proper places. Primaries, secondaries, tertials, and scapulars should all be properly aligned and placed before the bird is finally laid on its back onto the pinning board.

21. Pin skin out in its final form to dry. Pinning on drying boards (I use styrofoam) is preferable to the traditional wrapping in a thin layer of cotton or gauze, because it is easier to learn and the preparator has full control and access to most of the specimen (Fig. 11). The wings should be on the back, with the wingtips under the tail (i.e., lying on its dorsal surface); I hold it all thus with one hand until the first four to six pins are in place. I begin pinning by crossing a pair of pins over the end of the stick near the tip of the tail and another pair over base of the bill to anchor the whole bird, then pin the wings in place. Pins are then placed wherever required to obtain a smooth, symmetrical form. The whole specimen should be symmetrical and well preened; I usually have a narrow pair of forceps in hand for this during final pinning. At times, pinning a thin piece of cotton over part of the venter helps to smooth it (especially in fluid preserved birds), and wrapping the whole in gauze after a day of drying can smooth the contour of the whole bird. The drying skin can be unpinned or unwrapped each day to be checked and adjusted until dry. Large birds usually require a combination of pins and gauze, and should be rotated almost daily to allow complete drying and to prevent mold and rot. See below for more details on drying specimens.

22. Remove the muscular outer sheath of the stomach by cutting it

open longitudinally and peeling out the inner sac, in which the contents lie. Open this sac halfway without spilling the contents and put the whole thing, contents and all, into a small vial. Fill vial with 70% ethanol, tightly screw on the cap, and shake vigorously. Examine. Write a brief description of the contents in the catalogue. Some large birds lack a removable inner sac (e.g., some raptors, waterfowl). In some species a crop may be present that contains more intact food; preserve some or all of this material similarly.

23. Review the catalogue to be sure all data are recorded there. See Labelling and Field Catalogues above and Figure 1.

24. With all of the data now complete in the catalogue, and with clean hands, write labels: one for the skin, one for the skeleton, and a small one for the stomach contents vial (Figs. 2 and 3). If skin and skeleton are going to be catalogued into the same collection, the skeleton label can be less detailed than the skin label (e.g., leaving the back largely blank except for mass, sex, number, and disposition). But the field number should be written twice on the skeleton label and care should be taken not to get body fluids or blood on it, or the affected parts may be eaten by dermestid larvae during processing. At the University of Alaska Museum we catalogue the skin first (when it has dried), directly from the complete label onto the computer. This computerizes all of the data associated with a specimen and enables the printing of a final label for the skeleton, whose original label is retained but is usually marred, stained, and perhaps even physically damaged from exposure to dermestids during the process of skeleton preparation.

25. Tie the labels properly (Fig. 3), and attach the skin label to the skin, tying it around both the leg (midway on tarsometatarsus) and the stick to add strength and prevent future damage through accidental pulling.

26. Prepare skeleton for drying. Remove eyes from sockets. Pop them and squeeze out the fluid. Put them into the body cavity. No skeleton is complete without the sclerotic ring, a ring of bone plates in the eye. Skin or at least open up the skin on the unfeathered portion of the leg and toes to facilitate dermestid access. After cleaning the bulk of the meat from the leg and wing bones, place these lengthwise into the body cavity (Fig. 12). Using a light cotton thread, and holding the head against the side of the carcass, firmly (but not too tightly) wrap up the skeleton, making sure the pieces in the body cavity are nestled well inside and their ends wrapped to keep them in place. Tie off the end, then loosely tie the skeleton label around the neck with clean hands, using forceps to insert the label thread around the now wrapped neck (Fig. 12). Hang whole to dry or place in a screened box in a well ventilated place safe from scavengers and generally free of insects. Watch drying skeletal material to prevent mold growth and insect infestations.

27. Put small label for stomach contents into vial with contents and alcohol after the ink has dried (Fig. 3). Be sure it is legible in the alcohol.

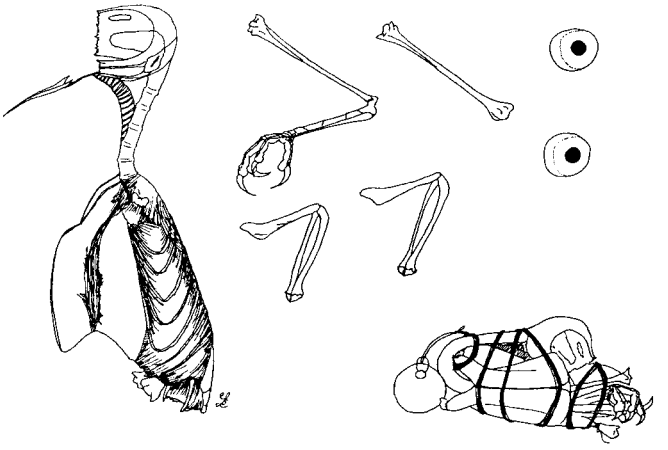

FIGURE 12. The carcass should be cleaned of large pieces of meat and internal organs. Eyes should be removed and the fluid squeezed out. Loose pieces, including eyes, should be put inside the body cavity and the whole well wrapped together in light thread, then labelled and dried. Second tibiotarsus and distal half of one radius and ulna can be left with skin to increase strength and to leave one wrist intact.

I also write the field number (twice) and species in indelible marker on the vial cap.

SEXING, AGING, FAT, AND SPECIMEN CARE

Determining sex.—Properly sexing each specimen is important. Sometimes it is impossible because a bird is too decomposed, or has been too badly shot, but it is generally possible, and a specimen of known sex is much more valuable to science than one whose sex is unknown or has been inferred from external features (in species where this is possible). Direct examination of the gonads is the preferred way to sex every bird. In many species sex can be inferred from plumage (sexually dichromatic species) or from the presence of sex-specific incubation patches (some species), but these inferences give us little or no detail about the present reproductive state of the individual.

Care must be taken not to disturb the dorsal region of the body cavity near the adrenal glands and the proximal ends of the kidneys until sex has been determined. Most errors in sexing, and the greatest source of unsexed specimens, arise from careless and overly exuberant removal of organs from the body cavity when taking tissues and gutting the specimen. Both sexes have a pair of adrenal glands that lie against the dorsal surface of the body cavity at the anterior, proximal ends of the kidneys (Fig. 5). Finding the adrenals helps orient the search for the gonads and prevents mistaking adrenals for testes in females by maintaining familiarity with the variation that occurs in adrenal size and color. In most immature birds, the basal third of the left adrenal is usually overlaid by gonadal

tissue, whether the bird is male or female. A stereo microscope or a hand lens in good light is an asset when preparing autumn and early winter specimens.

When found, gonads should be measured and described. Measurements include the maximum length and width of the organ(s), and the diameter of the largest ovum in females. Descriptions can include such things as the state and size of developing ova or eggs, condition of the oviduct, degree of differentiation of the ova, and color of the testes. Usually only the left testis is measured (usually being the largest), but care should be taken to observe both to be certain the bird is male. Most avian females have only one ovary (the left), but some (e.g., Falconiformes) have two. Figure 5 represents a nonbreeding adult of each sex.

Sexing adults during the breeding season is easily done because the gonads are usually very large. During the nonbreeding season, however, gonads can often be difficult to find because they are at their smallest. This is especially true in first year birds, whose gonadal tissues have often not yet become fully developed. The ovaries of young females are often not yet granulated with ova, and the testes of young males may not yet be opaque. First year autumn Parulidae, for example, can be quite difficult to sex unless very fresh or frozen quickly after death. When gonads are difficult to find, rinsing the body cavity in cool water (after skinning) can help, as can soaking the carcass in a bowl of water in a refrigerator for an hour or more. This usually causes the gonadal tissue to become a different color than kidney and adrenal tissues, making them easier to discern.

External indicators of reproductive condition should be noted with sex in the catalogue and on the label. Presence and/or condition of cloacal protuberances and incubation patches (sometimes inappropriately called ''brood'' patches, after their secondary function) are normally recorded. McCabe (1943) and Foster and Cannell (1990, and references therein) described avian reproductive conditions in greater detail.

Aging by skull ossification.—Skull ossification is a useful aging characteristic in most passerines and some other birds. As these birds get older (usually in the first year), the cranium gradually changes from a single bone layer to two layers. The two-layered cranium develops along a front, and the layers are separated by small bony pillars. As the two layers become fully separated and ossified along the developing front, the small space between the layers fills with air (becomes pneumatized). Many authors refer to this entire process as ''pneumatization,'' which is inappropriate. Ossification precedes pneumatization, and, moreover, natural stresses to the crania of wild birds frequently cause unpneumatized regions to develop in fully ossified adult birds (pers. obs.; see also figures in Klem 1990a). In hand-held living birds it is the contrast caused between pneumatized and unpneumatized regions of the skull (observed through the skin) that enables the bander to determine the stage of ossification and pneumatization. Ossified, pneumatized areas are pale; unossified regions are darker because brain tissue and fluid lie directly against the

inner side of the single layer of developing cranial bone. Unpneumatized areas are also darker, because fluid lies against the outermost layer of the cranium (whether there are one or two layers). The contrast between ossified and unossified areas is also easy to see while skinning, and the best time to record the condition of ossification is usually when the skin is first peeled from the head.

Unossified regions of the skull usually occur in bilateral symmetry, and are readily discernible when large. Unossified areas are always unpneumatized, but the opposite is not true. When asymmetrical unpneumatized areas are seen, look very closely to determine the actual state of ossification: observe the distribution of the small points caused by the little bony pillars separating the two cranial layers. Small, roughly symmetric unpneumatized areas (often termed ''windows'') should also be carefully examined for bone growth; they may represent the final stages of both processes. Good light and a hand lens or stereo microscope can be helpful in close examination. The state of ossification should be recorded as an estimate of the percentage of the cranium in which bone growth is complete, stating whether the skull is ossified, or, if not, approximately how much is ossified (e.g., 5%, 10%, 25%, 50%, 75%, 90%, or 95% oss.). The degree of ossification is important, particularly in species where the entire process takes more than a year. Ossification pattern is often sketched in catalogues and on labels, but this information is not readily computerized and is retrievable from the partial skeleton.

One advantage of the preparation method outlined here is that the skull is available for later examination. In birds that have died of head trauma (and in fluid preserved specimens) it can be impossible to determine stage of ossification until the skeleton has been fully prepared. Careful study of skulls to verify age following skeletal preparation has also proven very important in my own studies in which knowing the age of a bird is critical, especially in many species of Tyrannidae and Turdidae, wherein complete ossification is often not attained in the animal's first year of life.

Determining the age of a bird, whether from plumage stage or from the degree of ossification, often requires experience. During and immediately after the breeding season ''ad.'' for adult, ''juv.'' for individuals in juvenal plumage, and ''im.'' for individuals in first basic plumage with unossified crania are sufficiently simple, generic descriptors that fit most passerines. If in doubt, don't write anything except skull and gonad conditions, which can help subsequent investigators determine age more accurately. I use some bird banding age categories because they are more refined than traditional museum aging descriptors (Department of the Interior 1981, 1991), but they require some care in their use. Briefly, after ''nst'' for nestling and ''juv'' for individuals in juvenal plumage, the age categories I regularly use from banding literature are: ''HY'' (hatch year) for individuals still in the calendar year of their hatching; ''SY'' for individuals in their second calendar year; and ''TY'' for individuals in their third calendar year. In many species and individuals it is not possible to

determine age after the HY period, and in these cases they are denoted as ''AHY'' for after hatch year. After second year (ASY) and after third year (ATY) categories are also possibilities. The peer reviewed literature should be consulted for detailed aging criteria on a species-by-species basis.

The bursa and its measurement.—In most nonpasseriformes skull ossification is not particularly useful for aging. In some groups, such as Anseriformes, Galliformes, and Charadriiformes, the size of the bursa of Fabricius can be useful in distinguishing first-year from older birds (Hochbaum 1942, Kirkpatrick 1944, McNeil and Burton 1972). The bursa is an ephemeral, pouch-like organ that lies dorsally above the cloaca inside the body cavity (see Proctor and Lynch 1993). In game birds it opens into the cloaca and its depth is measurable in the living bird, but in shorebirds it is not so accessible. The length and width of the bursa is readily measured in preparation, however, and outside of game species it is an underutilized method of aging. Care must be taken in these taxa when cutting the spine above the tail not to damage the bursal region. Do not confuse the bursa with the caeca, which are paired and occur more proximally along the intestine.

Fat.—The amount of fat that a bird is carrying is important information best recorded after the bird is skinned. A six-level scale is generally used: none, very light, light, moderate, heavy, and very heavy. With experience, these divisions, although representing a continuum, become fairly obvious. They are based on the amount of fat found in the feather tracts, furculum, abdomen, intestines, and on the skin. Fat is assessed more thoroughly in skinned specimens than banded birds (contra Foster and Cannell 1990), although a six-level scale may be used in both. McCabe (1943) and Foster and Cannell (1990) both described a six-level fat scale, but their descriptions differ considerably, primarily because the latter was developed from passerine banding literature.

Briefly, ''none'' is recorded when little or no fat exists anywhere on the bird (even a starving bird can retain tiny amounts of fat in the abdominal cavity and on the dorsal tract). Very light fat is indicated by the presence of some fat in the dorsal tract and a trace in the furcular area (usually adheres to skin). Light fat adds depth to the former and some to the abdominal cavity. Moderate fat continues adding depth to the dorsum and furculum, and includes significant abdominal fat (inside and out) as well as small plates or pads of fat on the sides and elsewhere on the skin. Heavy fat is used when all of the feather tracts and much of the skin is covered with heavy pads of fat and large deposits are present in the abdominal cavity. At very heavy fat levels the body is entirely encased in thick fat, as are the intestines; fleshing becomes difficult because the skin, being stretched and greasy, is thinner and weaker.

Drying specimens.—Exposure of the pinned or lightly wrapped skin to air circulation is crucial until it is completely dry. A specimen is usually dry when its toes are no longer flexible. In warm and dry environments, drying specimens is usually not a problem; small birds can be fully dried

in as little as a day. Drying is aided considerably by moving air, and an electric fan is a superb tool for assisting the process, especially with large birds. Drying specimens is often a challenge under humid, wet, or cold conditions. Screen-bottomed racks over a stove set to give off a low heat are very effective; warm, dry air passes over the specimens from below. Care must be taken to keep heat levels low and to avoid exposure of specimens to smoke or flames. Putting skins in direct sunlight should be avoided; it causes plumage fading. However, using strong sunlight on a shade under which the birds are placed with adequate ventilation is effective because it provides heat. A refrigerator will slowly dry a specimen, and provides a good start on the drying process. It will also kill ants in tropical environments. Rotate large birds periodically during drying.

Drying skeletons is as challenging as drying skins under poor conditions, but the process can be assisted by immersing the thread-wrapped and labelled skeleton in 95% alcohol for a short time, then hanging it to dry. In all cases, dry skeletons slowly or the long bones may permanently warp. This is a problem when artificial heat is used.

Pests, and care of prepared specimens.—Arsenic, the effective poison and old museum standby, is no longer considered safe for human exposure. Even borax, a commonly used natural soap and mild insect repellent, has been eliminated from use in bird skin preparation because it has been implicated in accelerated foxing (color changes) of plumage (e.g., Phillips 1991:xliii; but not without some disagreement, K. C. Parkes, pers. comm.). Consequently, we use no chemicals when preparing bird skins. Borax does remain effective, however, when dusted on drying skeletons to aid drying and to provide a mild insect deterrent (D. Causey). Such skeletons must be well rinsed before putting them in a dermestid colony.

Keeping insects from specimens as they dry and before they are safely transported to a museum collection is an obligation and challenge. When completely dried, unpinned or unwrapped, and labelled, skins should be placed in a dark place where they are not susceptible to insect pests. Ant repellent or poison regularly sprayed or dusted onto pinning boards (*not* onto the birds) helps during the drying process. Paradichlorobenzene (PDB) crystals, a preferred form of ''moth balls'' still readily available commercially, will temporarily prevent insect incursions following drying and temporary packing when it is sprinkled liberally in a field case or specimen packing box (cardboard boxes work well). Specimens should be regularly inspected for insect activity. Personal exposure to the really effective insect-deterring chemicals should be minimized. For reasons of human health and skin specimen longevity, most museums now use no chemicals on the skins themselves, and also minimize use of insecticides and repellents.

Cryofumigation has become a popular method of ridding specimens of pests. Freezing specimens at −40 C for at least 48 h appears to kill all insect pests. Higher freezing temperatures (normal freezers rarely get below −20 C) are not completely effective, but insect activity is usually completely retarded. High concentrations of PDB probably kill and at least

severely retard the development of most insects. Together, two weeks at −20 C and a month in a case with a high concentration of PDB have proven an effective and legal method of fumigation. Every specimen coming into a collection or specimen case should be put through a fumigation process first. Insect damage is irreversible, can be rapid, and can also spread.

Skeletons.—The method outlined above generates labelled, roughed-out, partial skeletons. Upon drying these are ready to go to a dermestid colony for cleaning by beetle larvae, and subsequently the cleaning may be finished through cold water maceration. Matthiesen (1989) provided excellent details of the complete skeletonization process. If the further processing of dried, roughed-out skeletons into clean, museum-quality skeletal specimens is not deemed cost effective or appropriate, be aware that several museums will accept the donation of these specimens (properly labelled, of course) and complete the preparation process.

Speed.—We want to produce a high quality specimen in as short a time as possible. The quest for speed should never compromise the quality of the final product, however. Useful specimens with complete and legible data are, after all, the point. Blake (1949), noted for his preparation speed, recommended maintaining an orderly work table, placing instruments in a logical and consistent order, avoiding uncertain or unnecessary movements by focusing on the job, and keeping close track of time while practicing to increase speed and efficiency. Although most of his methods are outdated, these suggestions remain effective. Keeping the number of tools used to a minimum also helps.

With changes in preparation methods and data quality standards, the time required to prepare specimens has increased. Coues (1874:71), using primitive methods that retained neither skeletal material nor tissues, and which produced skins often damaged through even normal use, considered 15 min per skin in small birds to be good work. With practice, the method outlined here requires 40–90 min per specimen in passerines, everything included (each specimen being preserved in five pieces with associated complete and partial labels). It is washing and drying that pushes the time required to the upper limits. On average I can make up one specimen per hour.

When in the field, collecting and preparing 10 specimens a day is excellent work. If preparation is delayed until return to the laboratory, however, 20–30 specimens or even more can be collected in a good field day.

Holes.—When started, care must be taken not to enlarge a hole. Unless very large, most holes do not need to be sewn closed. They can usually be hidden by a final pull of the skin and arrangement of the plumage, or sometimes by slightly understuffing the bird at the affected spot. When it is necessary to sew a hole closed, one must be careful not to allow stitches to pull feather bases awry, for it is difficult to get such feathers to lie smoothly again.

Places students tend to make errors.—Treating the plumage roughly during skinning. Don't brush feathers away against their grain; don't blow

indiscriminately to clear an area. Instead, move feathers gently and carefully away from incisions or exposed skin by pushing or brushing them with the grain. The kinder you treat the plumage during skinning, the easier it will be to finish with a good-looking skin.

Using the scalpel too much in skinning: you only need it to make the initial incision, disarticulate, cut meat, and cut eyelids. Otherwise, use thumbnails, pressing skin away from the carcass (do not pull the skin). Using the scalpel elsewhere results in holes.

Duplicating the bird's neck in building the cotton body, resulting in an ugly skin with its neck stretched out too far. In their normal posture, most birds carry the head close to the body by bending the neck rather severely. In living passerines, for example, the furcular area is usually filled with neck. Make the cotton body up close to the tip of the stick, leaving only enough space (or just slightly more) for the stick to go into the upper mandible (to the nostril) and the head to lie close to or against the cotton body. All of the neck feathers should be visible, but the neck should not be particularly outstretched. Long-necked species (e.g., many Scolopacidae) and those in the Picidae and Anseriformes do require at least some neck to achieve a decent looking skin.

Not using enough dust. Pour it on.

Feathers sticking out in the lower neck and shoulder areas. This is caused by stretching the neck skin, which is usually folded with the neck. Before the skin dries, bunch the neck skin as described in step 20 above.

Twisted feathers disrupt the ordinarily smooth surface of the feathers. Grab the offending feathers before the skin dries and twist them back to where they belong (try both directions).

Head: make eyes the same size as the original eyes or just slightly smaller; make the head cotton the same size or slightly larger than the original skull minus the eyes. When putting in eyes, push the cotton well into the head skin, stretching the eyes outwards on the skin.

Arrange leg so that it is oriented correctly and the claws do not protrude beyond limits of tail. They tend to catch on packing cotton and other specimens, leading to damage. Also arrange it so the tarsus (tarsometatarsus) is readily accessible to measurement: keep it in the plane of the pinning board with the proximal end just exposed.

Tie label around both the leg and the stick together to increase the strength and durability of the specimen. Leave one inch (2.5 cm) of doubled thread between specimen and label. Less distance makes it difficult to measure the tarsometatarsus, examine undertail coverts, or read both sides of the label. Less distance also exacerbates problems of labels getting tangled in claws. However, a longer thread between specimen and label causes the label to become tangled with other labels and specimens.

Be sure to clean out the tail properly!

Body form: use the carcass as a model for the cotton body, and strive for smoothness, symmetry, and proper form in the final specimen. The skin usually dries rapidly after being put inside right. Once the cotton body is inserted, the preparator often must work quickly to get everything

into proper place while the feathers and skin are still manipulable. In slowly drying birds one can inspect and rearrange things the next day.

Completely evert wings, so that skin around the ''wrist'' is not bunched or wrinkled. Before adding any cotton, extend wings and arrange all wing feathers, especially secondaries and tertials, into their proper order and positions.

Labels properly tied (knots and distances) and written. Accurately following the directions given is not difficult, and the proper result is evidence of professionalism.

VARIATIONS

Large heads.—The heads of Picidae and Anatidae are generally too large to be removed through the neck. In these species, an incision is made either in the back of the head or in the neck and throat, usually whichever side will not be facing upward in the final skin. This incision is most easily made from the inside as one is skinning the head, and should be sewn up from the inside as well to most easily keep feathers on the outside.

Long necks.—Birds with long necks should generally be made up with the neck folded back along the body to save space in packing and in the museum case. In these birds the neck can be made up only of cotton or of cotton and wire. The head should be attached to the body with firm stitches of heavy thread, leaving the bill accessible for measurement. These stitches can be placed through any convenient part of the head skin, or in some cases the nostrils.

Large birds.—Large birds (e.g., large *Buteos* or *Larus* and up) usually require more substantial stuffing than just cotton. Excelsior, also called tow or wood wool and available from taxidermy supply houses, should be used at least as a body core, with a light cotton wrapping. To make excelsior bodies very tight and compact, wet the excelsior with warm water prior to forming and wrapping with thread, and let the body dry overnight. Stuffing large birds also requires a lot of material, not always readily at hand. I have had to resort to making rather flat specimens of gulls in the field (using cardboard forms), and to using mosses, crushed leaves, and accumulated cotton scraps. L. Stejneger once used his socks to stuff a gull (D. Causey, pers. comm.), and K. Parkes (pers. comm.) has had to use newspaper and shredded blueprints. Try to use archival quality materials, and, as usual, use the carcass as a model for size and shape.

In large birds it can occasionally be difficult to both tie the wings and to get them to dry against the body without flopping down. In very long-winged birds (e.g., Procellariiformes), I often find it easier to leave internal threads tied to each ''wrist'' loose in the body cavity until final stuffing, and then tie them together within the bird, or foregoing internal thread entirely and stitching the wrists to the skin in the proper position from outside, using heavy thread. Whenever doing the latter, you must be sure not to place stitches so they inhibit measurement of individual flight feathers (an abysmal old practice used by some preparators).

Shmoos.—When skeletal material is favored over a skin comparable to historic material, more bones can be removed during preparation for preservation with the skeleton—including the complete skull. Bill-less skins are called ''shmoos'' or ''muppets'' in the museum community, and provide researchers with an essentially complete skeleton and a skin useful for plumage analyses. The biggest difference in preparation is to skin the head completely, carefully cutting the skin away from the base of the bill from the inside with a scalpel when that area is reached at the end of the skinning process. In some species, much of the bill itself can be skinned. The head of the skin must be reconstructed more carefully than in the usual skin. Use the same amount of cotton for eyes and skull replacement, but leave final arrangement until after the stick is in place and the mouth sewn up. Insert the stick and cotton body as usual, but make the stick project just a little beyond the mouth. (I usually do not use a sharpened stick for shmoos). Using close, symmetrical stitches and working the needle always from the inside out, sew the mouth shut around and to the stick. A shmoo can be made up with no bones at all, but duplication of whole bones with the skeleton comes at great cost to a useful skin and should rarely be worth it. Dickerman (1989) provided more details on shmoo variations.

Open wings.—In a traditional skin, the closed wing prevents detailed examination of primaries, secondaries, and the inner wing surface. In many species (e.g., Dendrocolaptidae, Scolopacidae, *Catharus*) important plumage patterns are thus hidden from view. Although it is possible to prepare a skin with one wing open, it makes the specimen fragile and difficult to handle, pack, and keep in the museum tray. When separated from the skin, however, open wings are useful and easy to accommodate.

Open wing collections may have begun as a means to save something of the skin from specimens otherwise made up as complete skeletons, or from hunter-taken gamebirds. They are easy to prepare, requiring no fleshing after the radius and ulna have been removed. Whether preparing open wings during complete skeletonization or as a by-product of the method outlined above, the methods are the same. The wing is taken off at the elbow by cutting the skin and disarticulating the bones. It is then skinned to the distal end of the radius and ulna. In small birds these bones are disarticulated and removed for the skeleton; in large birds skin out the hand bones as well. The wing is then put inside right and flight feathers and coverts carefully rearranged on the pinning board. The wing should be pinned out in a naturally outstretched position; examine the wing before removing it from the bird to see what this should be. Artists often find these preparations to be some of the most useful in a museum collection. Open wings are also helpful for studying molt, particularly if the preparator had the sense to note whether any present was symmetrical.

To complete a skin preparation when one wing has been removed, Spaw (1989) presented the useful ''button-stick'' method. Instead of tying the outer wings together as in step 10 above, one uses a short stick as the

second wing and uses the same distance (or a little less) when tying this stick (at its center) to the remaining wing. This stick is then placed through the hole where the second wing was removed, there serving as a ''button,'' giving firmness and symmetry to the remaining wing and the complete specimen. With practice, a skin made up without one wing can have perfect form and be difficult to separate from an ordinary skin without turning it over.

Flat skins.—The traditional round skin is most comparable to historic preparations, but may not always be desirable or possible to prepare. For example, when processing a large number of specimens is necessary and the job can't be done effectively given the time required by the method outlined above, much time can be saved by preparing a flat skin and a partial skeleton instead. Similarly, a series of large birds takes up less space and can be prepared more quickly and effectively as flat skins and skeletons.

The method I use is to make an incision on the left side of the bird from the left side of the mandible, under the left wing, to the distal end of the left tibiotarsus. The shoulder is disarticulated first, then the skin is removed from the left leg and the bird is skinned nearly the same as usual, except that the right leg stays with the skin (the cotton around the right tibiotarsus provides strength to keep the leg attached). The right wing is removed at the elbow after skinning, and a cotton eye is inserted into the right eye. The bill is closed as usual with pin and thread, and the bird is pinned out skin-side down with the right leg cocked along the bottom of the specimen and the left wing, folded, pointed backward along the top side of the flat skin. The wing that was removed is pinned out as an open wing. A single label is threaded through the skin of the elbow area of the open wing and attached to the flat skin in a loose loop through the right wing hole. Storing these specimens in archival plastic sheets (photo album pages for small birds) maintains their integrity and prevents damage. All standard skin measurements can be taken, and colorimetric readings can be made as well, provided care is taken in plumage arrangement.

In small birds this preparation can be completed in 30 min or even less. It is particularly effective in processing partially freeze-dried specimens, when an aesthetic round skin is unlikely anyway. Spaw (1989) and Garrett (1989) presented other methods of producing flat skins, but, unlike the method presented here, neither produces a specimen that provides all the data obtainable from a round skin.

FINAL DISPOSITION

Scientific specimens should be deposited in a collection at an institution having a demonstrated long term commitment to specimen care and use. Such collections are most likely to be successful as long term repositories, and the specimens there will be most accessible to present and future researchers. Specimens are routinely loaned and borrowed among institutions through the mails, so a specimen's geographic location is of-

ten not particularly important. When deciding where to deposit specimens, considerations should include: whether the institution has permanent staff dedicated to collection care and use; the permits held by the institution (e.g., CITES, USFWS, USDA-APHIS); whether their collection is computerized and accessible over the internet (enhances accessibility to the research community); whether agreements are extended to researchers if specimens are being deposited while still part of a research project; proximity to the researcher if specimens are still needed; and international accessibility (trustworthy postal services). Institutions cannot accept illegally taken specimens except under very special circumstances. There is no current directory of the world's bird collections. However, many museums (including the University of Alaska Museum) will accept legal, prepared, data-bearing specimens for long term archiving, making them accessible to the world research community in agreement with the depositing individual. Getting specimens into proper long term archiving facilities in a timely manner is very important. Damages caused by insects, mold, light, etc. are considerable and irreversible.

Birds are packed for transportation or shipping in wooden or double-walled cardboard boxes. Specimens are usually wrapped in tissue paper to prevent cotton from catching on them, and are then packed densely and completely surrounded by heavy cotton. Boxes are packed full and with slight pressure so that there will be no settling or shifting of contents. Shipping labels should state, at a minimum, that the contents are scientific specimens of no commercial value. A complete inventory of the contents and copies of associated permits should be placed in an envelope on the outside of the package for any international shipping. Boxes of specimens are usually sent insured against loss.

CONCLUSION

There are many ways to prepare bird specimens. The general method outlined here has been developed from a lot of experience. It seems to maximize the usefulness of every bird prepared while not overly compromising the number of preparations one can perform. Skin-only or skeleton-only preparations can be finished faster, and may be preferable in special cases. However, sample sizes and overall specimen quality are greatly enhanced when specimens are prepared as combination preps. I have not considered many types of specialty preparations used daily by research specialists—fluid preservation (''pickling'') and the preparation of eggs and nests, for example. Persons wishing to learn more of these and other methods should begin with the excellent information assembled by Rogers et al. (1989) and Rogers and Wood (1989).

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APPENDIX

Sources of Supplies and Services

These sources are not meant to be endorsements; they are simply places with which I have done business in recent years. All are in U.S.A., but many deal internationally.

Skinning implements, cryovials.—1) VWR Scientific Products; many locations. Tel. 1-800-932-5000. 2) Fisher Scientific, Headquarters Pittsburgh, 711 Forbes Avenue, Pittsburgh, Pennsylvania 15219-4785. Tel. 1-800-388-8355.

Vernier calipers.—Forestry Suppliers, Inc. P.O. Box 8397, Jackson, Mississippi 39284-8397. Tel. 1-800-647-5368.

Stomach content vials.—Perfector Scientific, Inc., P.O. Box 91, Atascadero, California 93423. Tel. 805-466-8497 (Securi-Vial, 7 ml, #2142).

Cotton.—1) Henry Schein, Inc., 255 Vista Boulevard, Sparks, Nevada 89434 (KenVet KV 2287, non-sterile veterinary cotton in 1 lb. rolls). 2) Custom Hospital Products, Inc., 2623 S.E. Raymond, Portland, Oregon 97202. Tel. 503-231-9663 (70% non-sterile, non-absorbent cotton, 30% polyester fiber, ''Custom Cotton with Polyester'').

Label and heavy sewing thread.—Coats & Clark, Consumer Services, P.O. Box 12229, Greenville, South Carolina 29612. Tel. 1-800-648-1479 (#220, color 1).

Cob flour (dust).—Mt. Pulaski Products, Inc., P.O. Box 100, 904 N Vine, Mt. Pulaski, Illinois 62548. Tel. 217-792-3211. (#6 corn cob flour)

Mist nets.—1) AFO Mist Nets, Manomet Bird Observatory, Box 1770, Manomet, Massachusetts 02345. 2) Avinet, Inc. P.O. Box 1103, Dryden, New York 13053-1103. Tel. 1-800-340-6387.

Auxiliary barrels.—Robinson's Gun & Tackle, 855 Street Road, Southampton, Pennsylvania 18966. Tel. 215-357-7381.

12 shot.—Murmur Corporation, P.O. Box 224566, Dallas, Texas 75222. Tel. 214-630-5400.

Reloading supplies.—1) Gamaliel Shooting Supply, 1525 Fountain Run Road, P.O. Box 156, Gamaliel, Kentucky 42140. Tel. 502-457-2825 or 2830. 2) Graf & Sons, RR 3, Highway 54 South, Mexico, Missouri 65265. Tel. 573-581-2266.

High quality hand binding for notes and catalogues.—The Mount Pleasant Bookbinders, HC 71, Box 38-B, Augusta, West Virginia 26704.

Institutional binding for notes and catalogues.—1) University of Minnesota Bindery, Room 180 PSB, 2818 Como Avenue SE, Minneapolis, Minnesota 55414. Tel. 612-626-1516. 2) Page Book Binding, P.O. Box 187, 1891 Trumansburg Rd., Jacksonville, New York 14854. Tel. 607-387-4387.