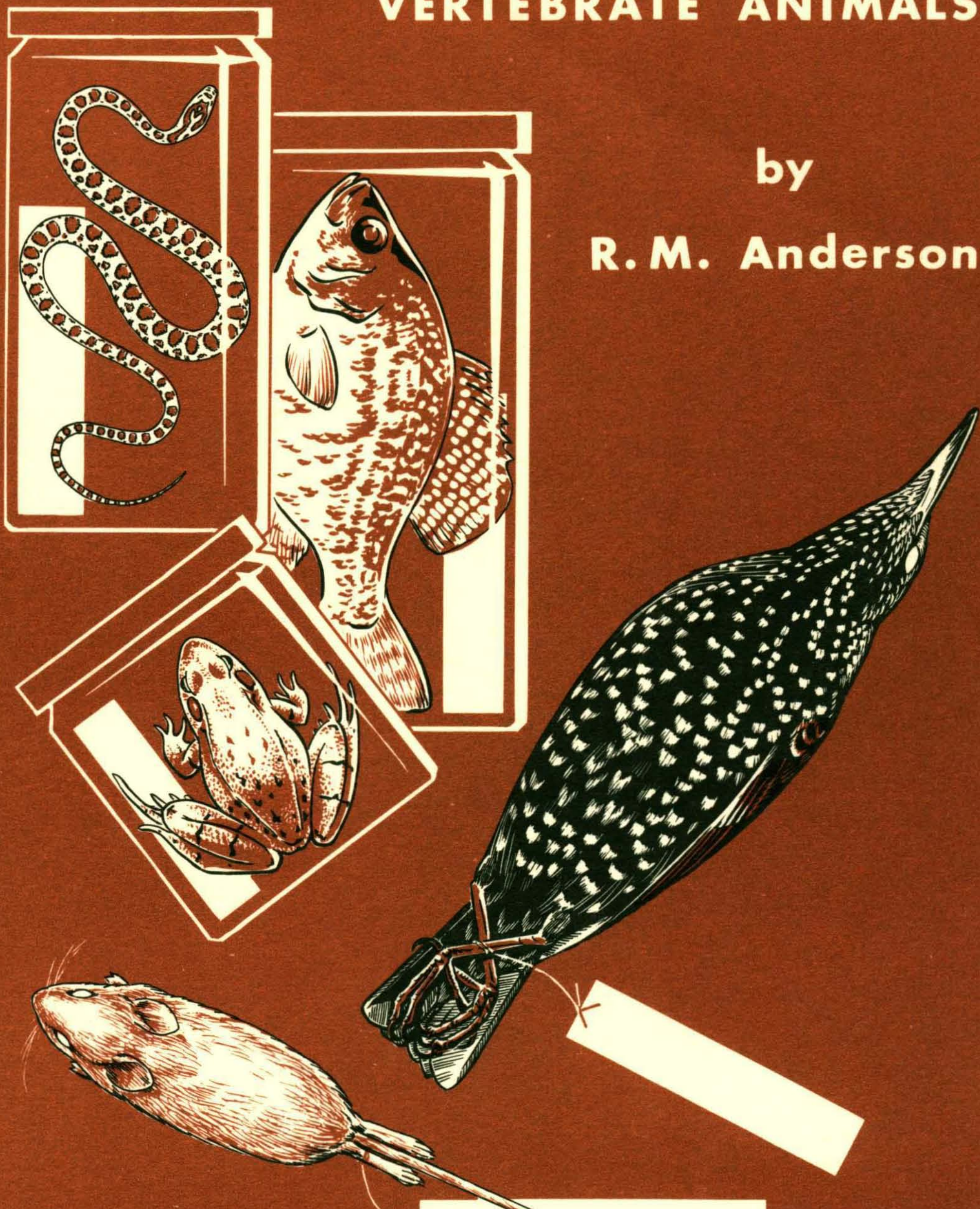
METHODS OF COLLECTING AND PRESERVING VERTEBRATE ANIMALS

by

R. M. Anderson



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BY
Rudolph Martin Anderson

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PREFACE

This first edition of this book was prepared in 1932 to fill a long continued demand from different classes of persons in various parts of Canada for information as to how to skin or preserve mammals, birds, fishes, and other animals. It is hoped the book will stimulate an interest in the National Museum prospective collectors. For the benefit of trappers, travellers in new regions, and others, information regarding the finding and catching of specimens is also given. Anybody who desires to collect specimens of animals of any kind is advised to read the whole of the introductory chapter before attempting work. There are many methods, but experience has shown that some do not give permanent results and, therefore, the author has given only those methods that have been found practicable in his experience, and in the revision has cited several later developments and improvements.

The manner of making up small mammal skins, and to some extent bird skins, was demonstrated in the laboratories of the National Museum of Canada by Mr. Charles H. Young, collector-preparator specialist, and all drawings were made by Mr. Claude E. Johnson, artist of the Division of Biology. Mr. Clyde L. Patch, chief taxidermist and herpetologist of the National Museum, made a number of helpful suggestions in the preparation of the second edition.

Major Allan Brooks, D.S.O., of Okanagan Landing, and Mr. Hamilton M. Laing, of Comox, British Columbia, are to be thanked for reading and criticizing parts of the manuscript, and for helpful suggestions. Thanks are also due to Messrs. H. E. Anthony and James P. Chapin of the American Museum of Natural History, New York; Mr. Herbert Lang, of Pretoria, South Africa; Dr. Glover M. Allen, of the Museum of Comparative Zoology, Harvard University; Mr. Wharton Huber, of the Academy of Natural Sciences of Philadelphia; Mr. Colin C. Sanborn of the Field Museum of Natural History, Chicago; Professor E. Raymond Hall, formerly of the Museum of Vertebrate Zoology, University of California, and later Director of Museum of Natural History, University of Kansas; Professor Homer R. Dill, Director of Museum of Natural History, University of Iowa; Mr. L. L. Snyder, Assistant Director of Royal Ontario Museum of Zoology, and Dr. C. H. D. Clarke, Fish and Wildlife Division, Dept. Lands and Forests, Ontario, Toronto; and Messrs. E. R. Kalmbach and Charles H. M. Barrett of the United States Fish and Wildlife Service, Washington, D.C., and particularly to Carl Akeley, "Father of modern taxidermy," for personal advice and communications not published; and to Dr. G. S. Whitby, formerly Director of Division of Chemistry, National Research Council, Ottawa (1937), for information on neutralizing formalin for preserving animal specimens.

The author is also under recent obligations to Mr. C. R. Twinn, Entomologist, in charge of Household and Medical Entomology, Division of Entomology, Science Service, Department of Agriculture, Ottawa, and to Mr. C. H. Bayley, Textile Research Laboratory, Division of Chemistry, National Research Council, Ottawa, for personal communications and recent publications regarding the use of "DDT" as an insecticide and repellent in various fields. Dr. Harrison F. Lewis, chief of Dominion Wildlife Service, Department of Mines and Resources, Ottawa, has supplied the latest information on collection permits for scientific purposes in Canada.

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METHODS OF COLLECTING AND PRESERVING VERTEBRATE ANIMALS

CHAPTER I

GENERAL PRINCIPLES OF ZOOLOGICAL COLLECTING

INTRODUCTION

The detailed and systematic study of animal life, which has advanced rapidly during recent years, necessitates extensive collections of specimens. The lower forms of life are most numerous, both in species and in individuals. Insect life is abundant in most parts of the world and the science of entomology now lists its species by hundreds of thousands. Improved vessels and new methods of deep-sea dredging have brought to light multitudes of new forms of marine life. Among the vertebrates, or back-boned animals, birds have always been evident to the most casual observer and the ornithology of civilized regions was scientifically studied at a comparatively early date. Collectors are continually bringing out new facts in regard to geographic distribution in all lands, and many new subspecies or geographic races are still being described from North America.

In field collecting, the first important thing is to catch the animal, and to do this it is helpful to know as much as possible about what animals may be expected to occur in a given area. Under References (page 145) are given titles and notes on a number of manuals and faunal lists which may be useful to a collector. Most of them will not be in small libraries, but a local naturalist will usually have some of them, and most good manuals and scientific papers give citations of other publications in the same field. Where possible, a collector should read every available publication on the area before he goes into a new field.

Mammalogy, or the study of mammals, though going far ahead in some lines, lagged for a long time behind other branches of systematic zoology. The anatomy, physiology, and pathology of the human race have been studied from remote antiquity and the results and technical names have been applied to the framework of the lower orders, but the lesser species were generally neglected. The principal reason for this backwardness is that, except for the larger mammals which are used for human food, or for their hides and fur, and others that are obvious pests, the majority of species of wild mammals are secretive in their habits and are seldom observed or taken unless vigorous and intelligent methods of trapping are used.

Miller (1929, 405)[1] attributes the recent development of mammalogy to two factors, an awakened interest in the nature and history of the life that now exists in the world, and the finding of a technique by means of which the study can be successfully carried on. The interest was aroused by the studies of Charles Darwin and the stimulating controversies that have

[1] The date following an author's name will enable the reader to find the complete bibliographic reference in the list of papers quoted at the end of this book.

never ceased to grow from his writings, and the technique was worked out by Dr. C. Hart Merriam, chief of the Biological Survey, United States Department of Agriculture, from 1885 to 1910.

The invertebrates (animals without a vertebral column) are more numerous and generally more easy to preserve than the vertebrates, but cannot be treated at length in this bulletin. However, as thorough field studies of vertebrates involve the collection and preservation of specimens of invertebrates, the student is advised to study a good textbook on ecological laboratory methods (Wight, University of Michigan, 1938, and others).

"Directions for Collecting and Preserving Insects" (McDunnough, 1928) may be obtained by application to the Dominion Entomologist, Science Service, Department of Agriculture, Ottawa. Small molluscs may be preserved in ethyl (grain) alcohol, but acids in some kinds of denatured alcohol (methyl hydrate) are apt to dissolve the calcium in the shells if they are immersed for any length of time. Large shells are usually cleaned of animal tissue and dried. Marine invertebrates of other orders, as well as land worms, slugs, etc., are generally preserved by immersion in alcohol or formalin, unless intended for histological or special biological investigations, when other methods may be used to prevent shrinkage of delicate tissues. The strength of the preservatives must be carefully regulated for the different classes of specimens (*See* section on "Preservatives, page 128).

The progress in the study of mammals may be judged by the fact that only about 150 species were known in North America a little over 100 years ago (Harlan, 1825), whereas the latest complete check-list (Miller, 1924) gives 2,554 forms inhabiting North America. Richardson (1829) listed 82 mammal species from British North America, and Tyrrell (1888) 137 species and varieties from Canada, whereas the latest Catalogue of Canadian Recent Mammals (Anderson, 1947) lists 594 species and subspecies of mammals known to occur north of the southern boundary of Canada, in addition to 2 introduced species and 49 of hypothetical occurrence.

The fact that a very large number of forms have been described and listed does not mean that our knowledge of them is complete in any case. Often a form has been described and named that appears to differ from all others, but only very few specimens may have been observed, and we are almost entirely in the dark concerning the extent of its range, its life history, economic value, or significance in the fauna of the country. All the larger mammals of Canada on account of their value as food or as fur producers, or because of their predatory habits, are without doubt known in a general way, and there is small probability of any new species being discovered. There are, however, local differences in some forms, due to geographical environment, that are worth studying, and few of our museums have really adequate numbers of specimens from the various regions. The differences in many cases are not readily apparent until specimens from different regions are brought together. With the advance of civilization and settlement most of the larger forms of animal life are rapidly disappearing all over the world, and many forms have become totally or nearly extinct within the memory of people now living. The necessity of obtaining adequate representatives of these rapidly disappearing animals for purposes of record and scientific study in the future is well recognized, but the haunts of the larger and rarer mammals are generally remote and comparatively inaccessible. The collecting of specimens becomes, therefore, both laborious and difficult. Few public museums have the resources for continuous and thorough work along these lines. Even the notable collections of such famous institutions as the British Museum and the Smithsonian Institution are largely due to the generous aid and contributions of private individuals outside of the technical staffs.

A considerable number of expeditions are made each year by sportsmen and big-game hunters into remote parts of the country, at considerable expense for equipment, transportation, and guides. Though many of the finer trophies are preserved and brought home, records are often lost and the trophies lose interest after the passing of the individual hunter. Many intelligent sportsmen, who enjoy the chase for its own sake, are coming to a realization of the scientific and economic waste involved, and by taking a little additional thought and care are providing the museums of their country with specimens that will be priceless memorials in time to come. For Canadians, or for sportsmen who enjoy the privilege of hunting in Canada, the National Museum of Canada is the legitimate repository of the zoological specimens they gather and which are not desired as personal trophies. Such acquisitions will be kept safely, the records preserved, and the specimens made available for scientific and educational purposes.

The small mammals of any district, with the exception of the commoner species of mice, squirrels, etc., are less well known than the larger mammals. Even the commonest forms may belong to local races of widely ranging species, and the particular forms inhabiting many regions are not definitely known. The smaller mammals, as a rule, are preyed upon by the larger carnivorous mammals and birds, and have developed secretive habits as a necessity for the preservation of their lives. The greater number of the small mammals are nocturnal and although their presence may be detected by different signs, such as the tracks in dust, mud, or snow, the animals themselves are rarely seen unless special methods are adopted for capturing them. It is safe to say that in any locality of varied topography there are to be found several species of small mammals whose very name and existence are unknown to more than one person in a thousand. Some small mammals are so secretive, and so very local in their habitats, that they may even elude for a long time the observation of a close student of natural history. For this reason there is a chance to discover new forms or species, and as so few districts have been studied thoroughly, there is an opportunity to obtain new records almost anywhere.

Though the study of the larger mammals, valuable commercially for flesh and skins, for purposes of sport, or for æsthetic reasons, is of obvious importance, and has been pursued in more or less sporadic ways from remote antiquity, the importance of the smaller, less conspicuous species is rapidly becoming recognized through the researches of modern science.

Many species of mammals that have no direct economic value as food, or for their fur or hides, are nevertheless often of enormous indirect importance. They must be recognized as beneficial or detrimental to the interests of man and if the latter, means must be taken to combat them. Ground squirrels, prairie dogs, pocket gophers, and other rodents have caused direct damage to grain fields, running into millions of dollars annually in some parts of the country. Rabbits, hares, and field mice frequently cause great damage to fruit trees and young forest trees. Coyotes, wolves, and mountain lions take a large toll of sheep, cattle, and horses, and large sums have been expended for bounties in combating them. House mice and rats are well-known destructive pests, and have been known to carry the germs of diseases to man and the domestic animals.

In addition to the obvious reactions on human enterprise of the species commonly classed as vermin, there are other relationships that are not so well known. Though it is commonly known to farmers, trappers, fur dealers, and naturalists that there are wide fluctuations in the numbers of various species of wild mammals and birds from time to time, the extent of such variations and their causes are not so well understood (Elton, 1924, Anderson, 1928, 1929, 1942). Many plagues of various species of field mice or voles have been recorded from North America, Europe, and Asia. In Arctic America lemmings and mice regularly reach the peak of abundance about every 4 years and this is followed by a rapid wiping out of nearly all the small rodents by disease. Arctic foxes and snowy owls increase in proportion to the lemmings and mice and fall away very soon after the depression in rodent life. The common snowshoe rabbit or varying hare has a very noticeable cycle of abundance, which reaches the peak about every 10 years. Muskrats and grouse show a similar but less-marked cycle, and various animals that prey on the others, such as lynx, red fox, mink, marten, etc., have parallel periods of abundance and scarcity. The small rodents have some local effect in settled regions, mainly in injury to agriculture, but in the wilderness their fluctuations have a tremendous effect upon the fur trade. As Elliot Coues once wrote: "They have one obvious part to play, that of turning grass into flesh, in order that carnivorous Goths and Vandals may subsist also, and in their turn proclaim, 'All flesh is grass' " (Jordan, 1929, p. 397).

The inter-relations between these different forms of life present still deeper and more obscure problems, which unfortunately have not received the attention they deserve. Pathologists have devoted most of their time to the study of diseases that affect mankind directly, and have discovered the relation of mosquitoes to yellow fever and malaria, of certain of the trypanosomes to sleeping sickness, of hook worms and other biological agents to other pathological conditions, and of rats and ground squirrels to the spread of bubonic plague and other diseases. It is also known that certain species of snails are secondary hosts to species of flukes that live in the bodies of various kinds of wild and domestic mammals, and that many kinds of parasitic worms and their allies infest the internal organs of other animals, but only a small number of their life histories have been thoroughly worked out. The most lowly species of mouse or shrew may be hosts to various internal and external parasites, or bear trypanosomes in their blood that may work havoc on themselves and on other species that feed on them or on the same range.

The problems of wild animal life are so varied and interlocking that the co-operative studies of systematic zoologists, parasitologists, pathologists, veterinarians, and ecologists, extending over a period of years, are essential to the solving of some of the important questions. Only the barest beginnings have been made in the scientific study of most of these questions as they apply to a few species. The key to some of these problems is only to be found by intensive and numerous prolonged observations in the field, and may be discovered by some farmer, trapper, game warden, or naturalist who is observant and reports his observations.

It is generally admitted by naturalists that species closely resembling each other often have quite different habits and unlike relations to other species. Different strains, as in plants, may not be subject to the same infections and their periodic fluctuations may be different. Observations on animal diseases, or on animal parasites, lose much of their value if there is no certainty of the host, or species afflicted. Consequently, there must be a certain amount of systematic taxonomic study before detailed observations can be made along other lines. Valuable observations of a general nature may be made without drawing the lines of differentiation too finely, but in general there must be sureness of the names of the animals before much of scientific value can be written about them (Anderson, 1920).

At this point it may be well to warn against incompetent or careless identification. A casual or "sight" identification by an experienced naturalist who is familiar with the mammals of the district is of more value than one by an amateur. However, many forms are so obscurely marked that there is great chance for confusion, and specimens need careful examination in hand. In case of doubt the specimen should be preserved and the determination verified by an expert.

Though certain methods of preparing specimens have been well tried, and are preferable, circumstances may arise when variations are necessary. An inexperienced collector may not remember the details, and not be able to go by the book, but if a skin is preserved with no external parts missing, and the skull, feet, and leg bones preserved, it is not important that the cuts be made in a strictly orthodox manner. These details are more important in handling short-haired or hairless tropical mammals, but fortunately most of our Canadian species are well haired or furred, and the cuts may be sewn up later without serious detriment to the specimen. Rare and valuable zoological specimens do not grow on bushes, and the collector has to take them when and under whatever conditions he can capture them. When they come, they may be in larger numbers than can be handled properly and emergency methods must be employed. It may be necessary to abbreviate the preliminary work and finish the details later. As long as the specimens are complete, they may usually be remade in a more satisfactory manner.

For persons who may wish to preserve specimens for naming or for record purposes, a simple method is described here (page 7). The method of preserving flat or cased skins will also be found very satisfactory for collectors who may happen to obtain occasional specimens when they have a small amount of equipment and limited time. Collectors in remote districts or on long packing trips will also find the method useful, particularly when time and opportunity are not convenient for drying the ordinary type of so-called "scientific skin" or "study skin". Persons who are making a large collection, and who are able to work under favourable conditions, will find that with some practice the conventional type of "skin" stuffed to approximate shape of the animal is easily and rapidly made, and more artistic and attractive. The flat skin, however, is of equal value for scientific purposes. In many ways, a good flat skin is superior to an over-stretched or poorly made stuffed skin. Study skins and flat skins may be mounted for exhibition at any later time. Specimens may be sent to the Director, National Museum of Canada, Natural History Branch, Ottawa, who will have them identified and returned if desired.

Specimens are desired from all localities. The only absolute proofs for range of species are specimens actually taken and identified. Collections have been made in many parts of Nova Scotia including Cape Breton Island, northeastern New Brunswick, Gaspe peninsula, the north shore of the Gulf of St. Lawrence and southern parts of Quebec, parts of Ontario, southwestern and northwestern Manitoba and the Churchill region on Hudson Bay, the prairie region of Saskatchewan, southern, central, southwestern, and northeastern Alberta, southern British Columbia and considerable areas on the coast and in the interior of the same province, southwestern and southeastern corners and Arctic coast of Yukon territory, the coastal region of the Northwest Territories, and some of the islands of the Arctic archipelago. These collections are, of course, not complete for any locality, they merely represent little oases of investigation, and work in nearby areas might bring remarkable additions to our fauna.

The areas that have received less attention are central, southern, and western New Brunswick, Prince Edward Island, New Quebec (Ungava Peninsula), parts of northern and western Ontario, southeastern and eastern Manitoba, northern half of Saskatchewan, northwestern Alberta, northeastern British Columbia, certain areas on the Pacific coast and islands and in central British Columbia, the interior of much of Vancouver Island, central Yukon territory, and large areas in the interior of the Northwest Territories (districts of Mackenzie and Keewatin). Within these areas at the present time may be found wide tracts of zoological *terra incognita* where the collector may work to his heart's content and be sure of adding interesting and valuable data on geographical distribution of species, life histories in a "state of nature," and the status and economic uses of the wild life.

In addition to specimens of every species obtainable, notes on abundance of the species in each locality are important, as well as information on the spread of a species into a new range or habitat. Records from localities that have been little studied are invaluable and before beginning active field work it is always helpful to read all the literature available on the fauna of a district. Records from the same place at intervals of time are useful to show changes in the species and in their relative abundance. Much valuable information may be obtained from old hunters, trappers, farmers, and market hunters; and such data from reliable "old-timers" is very important historically and if not jotted down will soon be lost.

The most common and characteristic species of a region are generally the most important, as they give the character of the local fauna, life zones, or association areas. Do not neglect the common species. Some collectors are prone to overlook them in the hunt for rarities. Some species are rare everywhere, but frequently a species which is rare in one place is common in another. The larger carnivorous species are naturally fewer than the animals that they prey upon.

A representative collection from each region is desirable as this is the only means by which it can be told where one form, subspecies, or geographical race blends into another. A beginner should, therefore, endeavour to collect specimens of everything that comes under his notice in his particular field. Where large numbers of specimens are available it may be necessary to choose, but a good rule is to preserve the first specimen of any species taken, as the collector may never get another of the same kind. A poor specimen is better than none, provided sufficient of the material is preserved to be identified and establish a record. Where it is absolutely necessary to discard some specimens because of lack of time to preserve them, a fair variety should be retained. Too close selection of "fine" or supposedly "typical" specimens may have a tendency to give a wrong impression of the average and may actually preserve abnormalities, a condition that often happened with the old school of collectors when a fine specimen of one male and of one female were the summit of the collector's ambition.

If collections are made in different localities, never assume that specimens from different places are duplicates of each other until they have been carefully studied and compared. Locality records in many cases are as important as species records. Many species vary under different geographic conditions and it is often difficult to detect the variation until the specimens are compared in the museum. A field mouse taken in Ontario may be the same as one taken in Manitoba, or may be of a different race, or an intergrade between two different forms; at any rate the possession of specimens is essential to determine the range accurately. In mountainous regions different races may be found at different altitudes only a few miles from each other. Do not neglect inconspicuous forms of life. The more showy and prevalent forms are apt to have been collected before, whereas the small and obscure specimens are more apt to be unknown.

Save specimens in different seasons if possible. Most mammals have different coats in summer and winter, the colour varying from dark brown to pure white, or the differences may consist only in length and thickness of hair. Intermediate stages are of value to show the condition of bleaching, or shedding of hair at different seasons. Specimens in worn and ragged coat may not be beautiful, but they may truthfully represent a stage in the life of the species. Also, a specimen may be taken largely on account of its skull or skeletal characters, and the skin, even if poor, may be a valuable check in the identification. Specimens of both sexes and of different ages or stages of development are desirable. The old idea of one male and one female being sufficient to represent a species in a museum has long been obsolete. Only a limited number of museum specimens are mounted for exhibition purposes and usually for such purposes perfect or "typical" specimens are selected, but other specimens are equally necessary for scientific study. As mounted specimens are apt to fade and deteriorate when exposed to light, additional specimens must be kept in reserve where natural conditions may be retained.

CARE OF SPECIMENS IN THE FIELD

Skins should be cleaned of all noticeable grease and adherent flesh, if possible. Sheets or chunks of flesh on a skin will prevent it from drying or prevent preservative from striking in. In a stuffed skin, even if the skin becomes cured, the flesh or fascia will contract and distort its shape. If the skins can be rushed to the taxidermist's shop or laboratory within a few days they may be heavily salted without thorough cleaning.

Sometimes a specimen will become so decomposed in hot weather that the hair slips and the skin becomes valueless. The skeleton of such a specimen may be preserved, and in any case the skull of most specimens is worth preserving. A scientific mammal specimen ordinarily consists of two separate parts—the skin and the skull, and, regardless of the market value of the skin, the skin and skull are of about equal scientific value. They should go together, and in some cases both are necessary for identification. Often either is valuable for separate study and is sufficient to substantiate a record. The skeleton may have the flesh roughly cut away and dried in the air, or if small, may be preserved in alcohol. When several specimens are available, it is a good plan to preserve one or two as skeletons; as well as alcoholic or formalin specimens for anatomical dissection.

Where it is impracticable to make any proper preparation of an unidentified specimen, it may be possible to preserve sufficient fragments for determination. For a bird, the wing, foot, tail, or head should be saved, or all of them if possible. For a mammal, the skull, a fragment of skin, tail, or foot should be saved, dried or in pickle.

TYPES AND TOPOTYPES

The type of a species or a subspecies is a single specimen from which the original description is made. There can be only one type of any given form.

Type specimens are highly valued in the scientific world and most of them are usually found in the larger museums where they serve as standards of comparison for specimens collected subsequently. Specimens other than the type used in making the original description are termed *cotypes* or *paratypes;* cotype when no type is designated, paratype when a type is indicated.

Topotypes, or specimens taken in the same locality as the type specimen of any species or subspecies, have a special scientific value for comparative purposes in identifying specimens, and collectors who are fortunate enough to be in the "type locality" of any Canadian species, are urged to preserve topotypes of the species found there. In the appendix to Miller's "List of North American Recent Mammals, 1923," pages 517-526, there is a list, classified by provinces, of one hundred and sixty-seven different localities in Canada from which two hundred and fifty-six different species and subspecies of mammals were originally described and named. Anderson's "Catalogue of Canadian Recent Mammals" (1947, pages 202-215) gives a classified list of two hundred and twenty-four type localities in the region covered by the Catalogue.

"Freaks" are not desired by a scientific natural history museum. Two-headed calves or other monstrosities are of little interest. Albino (white) and melanistic specimens (black, or with excess of dark colour pigment) are of some interest as a matter of record, but the specimens are of most interest to students of genetics and to fur-breeders who desire the living animal.

Specimens of either mammals or birds intended for a museum are much more desirable in the form of skins than as mounted specimens. Skins are more convenient for purposes of comparative study, are less breakable, and are much less bulky and, therefore, more easily stored. Great auks, Labrador ducks, passenger pigeons, Eskimo curlews, sea-otters, and other extinct or nearly vanished species are of interest in any shape, but most old mounted specimens, if not entirely worthless, are usually in such shape and pose that they do not fit into the general scheme of museum exhibition cases without being remounted. Mounted specimens are difficult to remount and unless virtually irreplaceable are hardly worth remounting.

TOOLS AND SUPPLIES

Tools. Flat skins of small mammals and even large ones may be prepared with a small pocket knife as the only tool. A good hunting knife or butcher's skinning knife is all that is really needed for the rough preparation of large mammals. The writer's preference is for a skinning knife not tempered too hard. A hard steel blade is easily broken, easily nicked, and difficult to resharpen. A 6- or 7-inch blade is most satisfactory, but the writer has found that the ordinary butcher knife of that length has too thin and light a blade for general field use, and it is better to buy a 9- or 10-inch blade, cut off the end, and file or grind to the desired shape. A blade bevelled on one side is easier sharpened with a piece of steel than if both sides are ground. If the butt end of the knife handle is tapered off to a broad, wedge-shaped end it is very useful in ripping the hide loose from the fasciæ and flesh during skinning operations.

Though an ardent and resourceful collector will never be deterred by the lack of an assortment of tools, the operations may be performed much more quickly, and usually better, if proper equipment is at hand. Not many tools are needed, but for small mammals and birds a small pair of scissors will be useful, in addition to the knife. A small pair of spring forceps or tweezers will be of much use, and almost necessary if finished study skins are to be made up. Other implements come in handy for special uses and may be added as needed, but for the beginner the following are sufficient:

- 1 small scalpel, or surgeon's dissecting knife.
- 1 large scalpel.
- 1 pair of small, straight-pointed scissors.
- 1 pair of slender forceps or tweezers.
- 1 pair of small pliers, with wire-cutter.
- 1 pair of dividers, for measuring specimens.
- 1 steel tape, 1 or 2 metres long, marked in millimetres and inches (marked to tenths if possible).

Other tools that will be found useful are butcher knives and heavy skinning knives for large mammals, a heavy pair of shears, a steel comb for dressing fur, a toothbrush for removing dirt and sawdust after drying skins, a flat file, a three-cornered file, and a good carborundum or oilstone. A scraper with toothed edge is useful for fleshing and removing the fascia or inner skin from mammals, and a soft rubber bulb-syringe or a hard rubber piston syringe is useful for removing the brains from small skulls.

An ordinary safety razor blade is used by some collectors for skinning and dissecting small specimens. A special handle may be obtained for holding certain brands of blades, but the blade may be held in the fingers or in a surgeon's artery-clip forceps. "Used" razor blades generally have a keener edge than the ordinary run of scalpels, and to avoid infection in pathological examinations may be discarded after using.

Skins may be scraped more easily on a tanner's beam, or the rounded surface of a smooth log, than on a flat surface. In scraping skins of small and thin-skinned mammals a small, round beam may be made of a piece of $\frac{3}{8}$-inch pipe covered with a piece of rubber hose set into a hole bored in the edge of a table. Large beams of the same type may be made of wood or metal covered with rubber tubing or a piece of automobile inner tube. Rowley (1925, page 123) recommends a scraper made of a sharp-toothed hacksaw blade 6 inches long set in a slot sawed in the edge of a flat piece of wood or aluminium shaped like an ordinary wooden lath. Hacksaw blades of different size in teeth must be used, as long teeth are apt to cut through thin skins. Rowley (1925, page 123) states that for fleshing and removing fat from small skins in the field, a light currier's knife handle fitted with a hacksaw blade, and used on an upright beam, is worth its weight in gold; and that professional trappers of fur-bearing animals could save much time by adopting this useful device, instead of cutting off fat and flesh with an ordinary skinning knife. For light work a satisfactory scraper can be made by flattening the bowl of a large iron mixing spoon, cutting the end squarely off, and sharpening and notching the edge with a file. Other articles that come in handy at times are found in the ordinary house or camp equipment. If the student wishes to go ahead and mount specimens, other tools will be found useful. Mounting mammals, however, is a skilled art which requires long practice, and cannot be treated in this booklet.

Preservatives

The preservative recommended as most satisfactory for the skins of small mammals and birds is a mixture, in about equal proportions by volume, of powdered arsenic (dry white arsenic trioxide, As_2O_3) and powdered borax (sodium tetraborate, $Na_2B_4O_7$). These ingredients are inexpensive and easily obtained. A pound of arsenic mixed with an equal volume of borax will serve an ordinary collector for a long time, and should be sufficient to preserve two or three hundred specimens of average size.[1] The exact proportions are not very important in practice. As the arsenic is the heavier and tends to settle to the bottom, the mixture should be frequently shaken up and mixed. This mixture is comparatively safe to use, as the borax is harmless and the arsenic dust is heavy and not easily inhaled. Arsenic is comparatively insoluble and is not easily absorbed by the skin, although if rubbed into an open cut it may make a small, festering sore, a purely local irritation. The mixture is poison, however, and should be kept conspicuously labelled, as mistaking white arsenic for baking powder has been known to cause unpleasant results. In the writer's experience, an overdose of arsenic taken by dogs or cats eating a thoroughly poisoned birdskin, merely acted as an emetic, without fatal consequences.

A mixture of powdered arsenic and powdered alum (aluminium sulphate, $Al_2(SO_4)_3$), in equal parts by volume or by weight, has for many years been a standard preservative for mammal and bird skins, and although

[1] Present regulations limit the purchase of arsenic. It may be obtained by authorized people for scientific purposes, but not by amateur taxidermists.

the "arsenic-and-alum" mixture has been more or less replaced by "arsenic and borax" for ordinary collecting purposes, the alum recipe is very useful in some cases.

The field collectors for the American Museum of Natural History, New York, who have done much work in the humid parts of South America, Asia, and central Africa, have generally adhered to the use of white arsenic (arsenic trioxide) or arsenic and alum in equal parts by volume (Chapin, 1923, page 6), but Mr. Chapin informed me in 1930 that he had recently been substituting borax for the alum almost exclusively. Alum has the disadvantage of hardening the skin somewhat, but a mixture of arsenic and alum may be very useful for very large birds in hot, moist climates. Chapin states that the arsenic is used for its permanent effect in preventing insects from eating the skin, rather than for any immediate action as a preservative, and that the arsenic should never be omitted. Anthony (1925, page 8) states that the skins of small mammals are best preserved by a mixture of arsenic and powdered alum (ammonium) the proportion about half and half by volume or by weight. Mr. Herbert Lang, who has had many years of experience in collecting mammals and birds in Africa, states that arsenic was necessary to prevent the attacks of insect pests, and advocates increasing the proportion of alum where the climate is very hot and moist.

Alum has an astringent or shrinking effect which is considered more or less detrimental to skins that are to be mounted later. The fur of certain species, as hair seals and squirrels with fur of certain shades of brown or yellow, are apt to have the colour of the hair changed to a deep yellow colour by either salt or alum, and the old-fashioned salt-and-alum bath for skins has largely gone out of use for that reason. Mr. A. H. Howell (1937, page 95) deprecates the use of salt and alum as it causes specimens to become noticeably altered in colour, and Professor E. Raymond Hall (1937, pages 359-360) warns against the method of salt and alum curing, stating that specimens of chipmunks, kangaroo rats, black bears, and mule deer may have colour of coat badly altered by its use. He states that if the salt-alum treatment is used, the least that should be done is to label the skins to clearly indicate departures from the conventional technique.

The small amount of alum used in touching up uncured spots will hardly be sufficient to affect the colour of the hair, but it may be well to attach a tag stating where alum has been used on the skin, so it can be scraped off before the skin is placed in the tanning solution.

The experience of the writer is that in the temperate parts of North America (including all of Canada) the use of alum is ordinarily unnecessary in curing specimens, and on account of its shrinking and hardening properties it is unsatisfactory for general use. However, alum is a very important agent for certain emergencies, and the field collector should always have a little on hand. In some cases when a valuable specimen has remained too long without being skinned, with the hair or feathers loose and about ready to slip off, a little alum in the preservative will "set" the hair and feather roots until the skin is dry. Sometimes in warm weather a mammal that is pretty far gone, with the skin turning green on the under parts, may be saved by quickly removing the skin and soaking it in a hot (but *not* scalding hot) solution. A little alum may be mixed with the fine sawdust that is used as an absorbent while skinning such a specimen. A little alum is also of great use for rubbing into soft spots and wrinkles of the skin while dry-curing large pelts in damp weather. Alum is much better than salt for touching-up uncured spots in fresh skins, as salt absorbs moisture from the air, causing the soft areas to spread. If salt is used on a skin, it must be thoroughly spread on the whole flesh surface of the skin.

The writer used arsenic-and-alum successfully for many years, but finally came to prefer arsenical soap (*See* formula below) when making up study skins. Seton (1921, 274) states that he came back to arsenical soap after trying other preservatives, because it is "safe," and best of all because the soap in it dissolves and absorbs some of the grease in the skin. It is by no means a substitute for the removal of fat by scraping and other mechanical means, but there is usually a small amount of grease in the skin that can not be raised by scraping, and when this grease is dissolved by soap solution, it may be absorbed by sawdust inside the skin.

The arsenical soap may be made as hard as desired and does not spill or leak. It may be kept in a wide-mouthed jar or tin, and when being used a sufficient amount on the top is softened with water and worked into a lather, which is applied to the skin with a brush. The liquid preservative touches and penetrates every part of the skin, softening the areas that may have dried out during the skinning and cleaning operations. Fine hardwood sawdust is immediately sifted over the whole of the moistened surface, and the surplus shaken off. The sawdust is not essential to the curing process, but the thin layer of sawdust keeps the poison from coming into contact with the fingers, and the feathers and hair of the specimen, and also aids in drying the skin. The job completed, any wet residue of the soap is poured off the receptacle and the container may be put away without danger of spilling. The remainder soon dries in the air without waste. Arsenical soap will hardly be mistaken at sight for anything edible, and the admixture of camphor (a slight insect repellent) is a warning to the sense of smell. Arsenical soap may be purchased from dealers in taxidermists' supplies, but it is easily and very cheaply made by Hornaday's formula (1892, 346):

White bar soap, soft rather than hard.....................	2 pounds
Powdered white arsenic..................................	2 "
Subcarbonate of potash (bicarbonate of potassium).......	6 ounces
Camphor	5 "
Alcohol	8 "

"The soap should be of best quality of laundry soap and of such composition that it can be reduced with water to any degree of thinness. Soap which becomes like jelly when melted will not answer and should never be used. Slice the soap and melt it in a small quantity of water over a slow fire, stirring sufficiently to prevent its burning. When melted, add the potash, and stir in the powdered arsenic. Next add the camphor, which should be dissolved in the alcohol at the beginning of the operation. Stir the mass thoroughly, boil it down to the consistence of thick molasses, and pour it into an earthen or wooden jar to cool or harden.[1] Stir it occasionally while cooling, to prevent the arsenic from settling at the bottom. When cold it should be like lard or butter. For use mix a small quantity with water until it resembles buttermilk and apply with a common paint brush."

Arsenic, borax, alum, and any of the dry preservatives may be kept in wide-mouthed bottles or wide-mouthed tins with friction-top or screw-top

[1]An ordinary glass fruit jar or wide-mouthed bottle may be used if the mixture is sufficiently cooled not to crack the bottle.

caps. Boxes or tins with loose lids should be avoided, as the powders are apt to sift out into the other luggage. Arsenical soap may be kept in a paste jar with two compartments—the soap in one compartment and a small brush in the other in which water may be placed when needed for use and poured out when finished. A very convenient method on packing trips is to carry the powders in double paraffined bags, made by putting one bag inside another bag. When in use, the tops of the bags may be rolled back to scoop the powder out, or the specimen may be dipped into the bag.

As some persons are peculiarly susceptible to local irritation of the skin when handling arsenical preparations, particularly when arsenic is used in quantity for long periods, various non-poisonous mixtures are coming into common use by collectors. These non-poisonous compounds preserve the specimens effectually from decay, but are less effective than arsenic in keeping insect pests away. As all specimens are unsafe from attacks of moths, dermestes, etc., except in far northern regions, unless stored in insect-proof cases, this disability is perhaps more apparent than real. The use of arsenic is not strictly necessary when a collector is working for a properly equipped museum where the specimens can be placed in insect-proof cases within a reasonable time. If fumigated on entrance such specimens will keep indefinitely without the use of poisonous preservatives. Some authorities do not consider arsenic a sure preservative against insect pests, but where arsenic-treated skins have spoiled the cause is usually imperfect and inadequate poisoning, particularly around the terminal bones of wings, feet, and beak of birds, and the toes of mammals. The writer has stored skins that were preserved with straight white arsenic in a case with large cracks open to moths and dermestes (bacon-beetles) for 25 years, and taken them out uninjured by insect pests, whereas other skins in the same case had been riddled by insect larvæ. The moral seems to be that collectors who do not have proper air-tight cases for permanent storage should use arsenic thoroughly, and plenty of it, in preserving their specimens. If used with ordinary care, arsenic offers no danger to the health of the collector unless he is constitutionally susceptible to its effects.

Dr. Glover M. Allen, of the Museum of Camparative Zoology, Harvard University, recommends the use of a mixture of sixteen parts saltpetre (potassium nitrate, KNO_3) to one part of alum as a preservative instead of arsenic. Some of the field collectors for the same museum have used this mixture with success in many lands. It seems to set the hair, does not liquefy in damp weather as salt does, and has the advantage of being non-poisonous.

Mr. Colin C. Sanborn informs the writer that the Field Museum of Natural History, Chicago, uses borax mixed with naphthaline crystals ($C_{10}H_{18}$) for preserving both mammal and bird skins, and the mixture has proved to be satisfactory on several extended field trips to South America. The borax and naphthaline mixture has also been used with good results by some of the collectors for the National Museum of Canada. The borax forms an effective preservative and has the advantage of dissolving readily if the skin has to be relaxed later. The naphthaline discourages blowflies from attacking skins that are drying, and probably repels mould and insect pests to some extent while the specimens are in transit from the field to the laboratory. A little fine hardwood sawdust mixed with the borax economizes borax, keeps it from becoming lumpy, and aids in drying the skin by absorbing dampness and the small residue of grease that is apt to remain on the skin. The borax and sawdust mixture is effective without the naphthaline. Too much reliance should not be placed on the naphthaline, because although it is a fairly effective deterrent to most pests when used in quantity, the small amount in a preserved skin will soon evaporate and the skins need to be kept permanently in an insect-proof case. Napthaline is the main constituent of the ordinary moth-ball, which may be used in cases of specimens when naphthaline flakes are not available, but the flakes are cheaper and being much more volatile are generally more effective.

Common salt is useful for temporary preservation, is obtainable anywhere, and is the best medium for preserving the fresh hides and scalps of large mammals. Salt may be used for preserving any kind of skin, and the finer the grain the better. Table salt is best for small animals, and ordinary barrel salt will do for large specimens. It should be applied plentifully and as soon as possible after skinning. A 2- or 3-pound container of one of the brands of table salt that will not become lumpy will preserve a considerable number of small specimens and may be poured out of a funnel without waste. Major Allan Brooks (in letter, 1935) advises that borax should never be used on mammal skins as it turns the fur red. He states that kangaroo rats (*Dipodomys*) from Death Valley, California, are a red race, but Mr. A. J. Van Rossem had shown him the year before that the same reddish type can be produced by using borax in the make-up of the skins of other races of the same genus. The writer was informed in 1937 that collectors for the Royal Ontario Museum of Zoology, Toronto, were using borax *only on bird skins*. Downing (1945) sums up experiments in the same institution relaxing and re-making skins of both mammals and birds that had been preserved with arsenic, arsenic-and-alum, alum, borax, and salt. His conclusion is that skins of many species of mammals, particularly the Sciuridae (squirrels) and other species with reddish or brown hair that have been relaxed and remade suffered changes in colour that renders them almost useless for taxonomic studies where colour is an essential character. Similar changes in colour were found to occur in plumages of remade specimens of several birds in different groups with reddish or brown colours. He states that the extent of the change in colour appears to depend on the length of time the hair is in a moist condition, and that it may be possible by rapidly relaxing, washing, and drying to avoid producing any colour change. This problem requires further investigation. In the meantime any specimen that is relaxed and made over or washed in the process of preparation should bear this information on the label.

An experiment checked by the writer on skin of red squirrel, freshly killed, and split down the back, one-half immersed in pure "soft" lake water all night, and dried, showed no perceptible difference in comparison with the half of the skin that was not wetted. It may be that borax or soap in water relaxing a skin, may remove some of the natural oil from the surface of the hair and make it more subject to change of colour in pigment cells. Chlorinated tap water may also have some effect in changing the colour.

Borax has not yet been shown to have any deleterious effect on the hairs of blackish or greyish colour, and its most injurious effects are shown when it comes in contact with damp fur of brown, reddish, or fawn shades. By far the greatest proportion of skins preserved are taken for scientific study purposes and do not need to be relaxed and made over. Borax is not an insect repellent, and as a well-cleaned skin really needs much less preservative than is generally used, and flat or cased skins will dry in the air without chemical aid, it seems desirable to put on less preservative on most skins. It does not penetrate the skin to any great extent, and if it seems necessary to make over a skin, it should be softened from the inside, and the preservative be kept as much as possible from contact with damp fur and feathers.

Although all the preservatives mentioned have some good qualities to recommend them, it is by no means necessary for the collector to keep a large variety of chemicals on hand. Most of the preservatives may be obtained in the ordinary chemist's shop or grocery, and the experienced collector will ultimately select the preservative that best suits his needs. It is, however, best for the collector to know as many as possible of the approved methods and preservatives, as he is apt to have occasion to prepare specimens at places where his favourite recipes are not available. All the preservatives listed are comparatively inexpensive, amounting to less than one-tenth of a cent for the average small specimen. Economy in use has, therefore, little to do with the initial cost, but is concerned chiefly with making supplies last through a long field trip on which they cannot be replenished. A workman or an artist may not be able to produce a prize specimen of handiwork without good tools of his trade, but an enthusiastic collector soon comes to realize that certain specimens have a great scientific value and must be preserved when they come to hand, even under the most discouraging circumstances. A competent field collector should be able to prepare anything from a field mouse to a moose with nothing but a jack-knife, if necessary.

Fumigation

When specimens are put away it is well to fumigate the inside of the case or box freely with naphthaline flakes or moth-balls. Paracide (paradichlorobenzene) flakes have similar insect-repelling qualities and though neither will kill insect larvæ in a skin that is actually infested they will generally discourage insects from entering. The efficacy of either depends largely on the tightness of the container, and cracks may be sealed by pasting strips of paper over them. Specimens may also be sealed up in strong paper bags or tied in fine cloth bags.

Stone brimstone (sulphur) broken into lumps and scattered in drawers or boxes where furs and woollen articles are stored, is said to be a good preventive against moths and has the advantage of being odourless. Like naphthaline, it cannot be considered a sure protection against insect pests, but will probably discourage them from entering fairly tight containers.

Where skins are actually infested with insect pests, carbon disulphide (bisulphide of carbon, CS_2) has been in general use as a disinfectant, as it is very volatile and the gas is heavier than air and works down through the specimens. As the gas does not always kill the eggs, the treatment should be repeated after an interval of a week or two to kill any larvæ that may have hatched out before they become large enough to lay any eggs. As the gas is explosive it should be used in a tight case and kept away from sparks or fire. More recently a more satisfactory agent has been discovered, a mixture of three parts by volume of ethylene dichloride ($C_2H_4Cl_2$) with one part of carbon tetrachloride ($ChCl_4$). Its advantages are "being highly toxic to insects, non-explosive, non-inflammable, non-toxic to man unless highly concentrated and breathed for protracted periods, non-injurious to specimens, inexpensive, and easily applied" (Leechman, 1931, 135-136).

Gassing methods are necessarily impracticable in the field, except in a small way where specimens may be placed in covered tin cans, etc. The methods of fumigating large specimens are treated in the following pages (page 74).

DDT as an Insecticide

DDT (dichloro-diphenyl-trichloro-ethane) is the most outstanding and the most widely used of the newer insecticides. The compound was discovered in 1874, and rediscovered as an insecticide about 65 years later, becoming extremely effective in combatting insect vectors of disease during the latter part of World War II. Its extraordinary effectiveness against many species and its prolonged residual action, its formulation, application, and the precautions that should attend its use have been well publicized in technical and popular articles (*See* References, Gibson and Twinn, Supplement by Twinn, 1946; Twinn, 1945, 1946, 1947; Twinn and Balch, 1946).

Twinn (1945) states:

"DDT is classed as a poison, but can be used safely if applied and handled with proper precautions, as outlined in the containers in which the product is sold. Contact of the skin with oil solutions should be avoided as much as possible, as DDT can be absorbed in this form. It is advisable to wash exposed parts of the person with soap and water after any considerable use of oil solutions or concentrated emulsions . . . DDT in any form should be kept out of foodstuffs, a recommendation which applies to any insecticide. In the dry form, such as in powder formulations, DDT is safe to handle, but persons using it should avoid inhaling or swallowing the dust. Tying a handkerchief across the nose and mouth is a good precaution.

"Insecticides offered for sale in Canada have to be registered in accordance with the provisions of the Pest Control Products Act. Under the regulations of this act the ingredients, guarantees, purposes of the product, directions for use, and the text of labels of such insecticide materials are reviewed by competent officials of the Department of Agriculture before applications for registration are accepted. Mention of this is made to assure the public that proprietary preparations containing the new insecticide DDT may be purchased and used with confidence, in accordance with the claims of the manufacturer or vendor shown on the labels of such products."

Mr. C. H. Bayley, Textile Research Laboratory, Division of Chemistry, National Research Council, Ottawa, writes (*in litt.*, 1947):

"It (DDT) is coming into wide use as an agent for protecting wool garments and fabrics from attack by the webbing clothes moth and the carpet beetle . . . The material is usually applied in the form of a solution in some organic solvent, either by spraying or dipping. However, it should be pointed out that in the case of specimens which may have become infected, it may be necessary to arrange for a fumigation of the entire specimen, using some toxic gas, since spraying the outside of the specimen would not kill the insects in the interior. In such cases a combination of fumigation and spraying should be effective . . . The toxicity of DDT when used in the quantities required to protect perishable specimens would not be expected to be appreciable. Actually DDT was used during the war for the impregnation of garments worn next to the skin for the purpose of controlling lice".

Filling and Wrapping Materials

Cotton batting is commonly employed for filling small skins of mammals and birds. It is economical and desirable to have two or three grades. For the smallest skins, as shrews, and for wrapping tails of mice, filling eye-cavities, etc., it is well to have a small roll of the finest grade of surgical cotton or jeweller's cotton. A half-pound roll will last a long time. For wrapping the legs, making bodies of small mammals and birds, and packing small, delicate specimens, it is important to use a good grade of cotton batting, without lumps, as lumpy cotton will cause a roughness in the made-up skin after it has dried. Cotton that can be peeled off in thin sheets is essential for wrapping bird skins, and it can be used again and again. For larger specimens and for packing around dried skins, a cheaper grade of cotton will do.

Fine tow makes a very good stuffing material for study skins, and if good quality tow with long, soft fibres is procurable, it may be substituted for cotton in most cases. Fine excelsior (stringy wood shavings) forms a very good filler for large bird skins, but is not very good for mammal skins, except for purely temporary purposes. Mammals larger than a small fox or woodchuck are generally best prepared in a flat state and subsequently put through a regular tanning process. Tanning, however, is a shop and laboratory process and need not be considered in the field. Oakum, dry moss, fine dry grass, or any soft vegetable material may be used for filling, but wool or other animal substances should be avoided as insects may attack them and destroy the specimen.

Mr. Charles H. M. Barrett, taxidermist of the U.S. Biological Survey, recommends carrying some finely chopped tow mixed with excelsior and fluffed up by placing the chopped material in a container that has a closed top. Compressed air is applied for a few moments under moderate pressure. This will fluff up the mass to an even consistency and it can then be pressed, rolled, or felted into shape. Mr. Barrett does not think it is better than cotton for making small bird skins, but in some instances it works well in small mammal skins. In making up large- and medium-sized bird skins it is useful in filling in parts like eye-sockets, sides, and root of tail. He uses a container made from a 5-pound lard tin, with a covering made of 8-ounce duck or canvas with a hole cut in the centre to receive the air hose. The hole is made round, sewn with buttonhole stitch until it makes a tight fit for the air hose. To make the canvas airtight it can be waxed or shellacked on the area covering the rim of the tin. The cover should be made large enough to overlap the rim of the tin for 2 or 3 inches and may be tied down with a piece of hard twisted cotton twine. The fluffed filling may be made in quantity and carried to the field in tins or boxes to be used as needed. If large quantities are needed it might be well to use a large container with a tight metal top with a piece of pipe extending about halfway down and projecting from the top about 2 inches for attachment of air hose or pipe.

Cheesecloth is the best material for wrapping large bird skins for drying, and the collector should take about 10 yards of cheesecloth on a field trip for wrapping skins as well as for keeping blowflies off specimens and fresh provisions. It also comes in handy for washing and wiping specimens, wrapping fish specimens, or any emergency in which a piece of soft cloth is needed.

Absorbents

Fine hardwood sawdust, such as is used by tanners in cleaning furs, is the best absorbent for use while skinning either mammals or birds. Maple or birch sawdust is preferable to oak or redwood, as the latter may cause stains if wet. Hardwood sawdust may be obtained in quantity from furniture factories or tanners. The very finest grades such as boxwood sawdust, which is favoured by furriers for "drumming" fine furs (prolonged shaking up in a revolving drum), are much like flour, and are not as good for field use as slightly coarser grades, which give a better grip and are easier to brush out of fur or feathers. The sawdust may be used over and over again, and the dirty and bloody parts thrown away each time. It is best kept in a waterproof bag, such as a paraffined "grub-bag", which can be tied tightly and the mouth rolled down when in use. Three to five pounds of sawdust will last for hundreds of small specimens. Use sawdust freely, putting on pinches or handfuls from the beginning of skinning operations to absorb any grease, blood, serum, or intestinal fluids. Professional trappers will find a little bag of sawdust an invaluable aid in skinning operations, as it helps keep the fur clean, and if the skin is lined with greasy fasciæ, a coating of sawdust gives enough grip that adherent matter may be more easily pulled or scraped off.

Pine or other softwood sawdust may be used, but is not as satisfactory as hardwood, being usually coarse and containing gummy and pitchy substances that detract from its absorbent qualities.

Fine cornmeal is about as good as sawdust for this purpose, and some collectors prefer it on bird skins, as fine sawdust has a tendency to stick in the feathers. The writer is accustomed to keep little bags of both sawdust and cornmeal on hand, using the cornmeal on bodies that are destined for the cooking pot. Sawdust is hard to remove from the flesh, and in the field it is often desirable to keep your specimen as well as eat it. When hungry-eyed comrades watch the skinning of scientific specimens, it is well to make concessions to the commissary.

Dry powdered clay, sand, dust, or wood ashes may be used for absorbent in emergency. Dry powdered whiting (purified calcium carbonate), such as that used as an ingredient in putty, or for cleaning silver and brass, is a useful absorbent on fresh shot wounds, and when dry may be crumbled and brushed off, taking away most of the stain. The writer usually carries a small bottle or can of either whiting or cornmeal in his pocket to absorb fresh blood from gun shot wounds. Ordinary wheat flour should not be used, as it forms a sticky paste. Plaster of Paris is much used by taxidermists for cleaning bird skins after washing, but it has the disadvantage of leaving a light powdery bloom on the feathers, particularly unsightly if they are of dark colour. Chapin (1929) recommends as the best material for cleaning feathers, a mixture of plaster of Paris and potato starch or potato flour (equal volumes), stating that it may be used in place of cornmeal throughout, and acts far more rapidly.

Cleaning Materials

Blood and grease should be removed as thoroughly as possible after the skin has been removed and before putting in the filling material. If blood is allowed to dry into the hair or feathers it is very hard to remove. Especially is it important to keep blood from touching the feathers. With most mammals, particularly the glossy, dark-haired species, fresh blood on the hair is not such a serious matter, as blood stains are usually removable by washing in cold water. Grease should be removed, as it will ooze out by capillary attraction, colour the hair or feathers a dirty yellow, gather dust, and ultimately "burn" the skin, causing it to disintegrate. The removal of grease is expedited by the addition of a pinch of washing soda (sal soda, sodium carbonate, Na_2CO_3) or a little soap to the basin of water. The hair may be sponged clean with a wet rag, using several waters if necessary, and the fur dried by application of a little dry sawdust, or by rolling the skin in sawdust, which may be shaken or beaten out. An excess of grease in the skin, hair, or feathers of a small specimen that is to be made up at once may be washed or sponged out with high-grade white gasoline, benzene, or carbon tetrachloride ($CHCl_4$). Carbon tetrachloride has the advantage of being non-inflammable. Acetone (C_2H_6O) alone or in combination with either of the above is an even better agent for sponging off blood or grease, but it is very volatile and both the liquid and the gas are very inflammable. Huber (1930) recommends a mixture of gasoline, alcohol, and turpentine (*See* page 104).[1]

Light-coloured or white animals, with soft, tubular hair, such as antelope, bighorn, and white sheep, should be kept free from blood if possible, as the hairs soak up the blood and in spite of repeated washings enough blood will remain to mark the spot. The white fox can be cleaned, but hares are more difficult. The ptarmigan in winter plumage is very difficult to clean perfectly, but white gulls and other water birds are, on the other hand, rather easy to renovate. Collectors for the Royal Ontario Museum of Zoology have recently been using powdered borax for absorbing blood on feathers of recently killed birds. When the borax has dried it may be brushed out with a toothbrush, usually leaving the feathers clean and fluffy.

Pins, Needles, and Thread

A paper or two of ordinary white pins (nickled brass) should be on hand for wrapping up bird skins, and may be used for pinning out mammal skins to dry, but for the latter purpose, sharp, black-headed steel pins or black steel (japanned) insect pins (No. 3 or larger) are much more satisfactory, as they are more slender, leave smaller holes in the skin, and do not spread the toes so much. They are, also, sharper and the fingers may become sore sticking many dull pins into a drying board. At least five hundred steel pins will be needed for active collecting, and they can be used again and again. A large sheet of flat cork, such as entomologists use, will be found very useful to pin out small mammals for drying. For field use, it is convenient to keep pins in small desk pin-holders.

A paper of assorted sewing needles, or lots of sizes 2 to 8, should be kept on hand, as needles are easily lost and are often broken in sewing skins. A few glover's needles (three-cornered) often are useful in sewing heavy skins. Two or three heavy sail needles and a curved bagging needle are handy for baling up heavy skins for shipment, and a ball or two of heavy cotton twine, coarse bagging twine, and a quantity of heavier manila cord should also be kept on hand for various purposes. A sailmaker's sewing palm will be found useful in sewing heavy skins, bags, repairing tents, pack saddles, and dog harness, etc.

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[1] Liquid or powdered detergents give excellent results provided *all* clotted blood has been removed from the feathers or hair. Rinse well in clear water, blot skin on dry cloth or paper towels, and dry it in an air-stream from an electric fan or a vacuum cleaner with blower attachment. Shake the skin while drying to make the feathers or fur fluffy.

Cotton thread Nos. 8, 25, 36, 40, 50, and 60 should be kept, as well as linen thread Nos. 40, 50, and 60. Two or three spools will be sufficient for sewing up several hundred small specimens, but it is well to have a few extra spools of the larger sizes for tying labels or wrapping up skeletons. Strong linen thread, No. 40, is preferable for tying small paper labels. A smaller size would perhaps have sufficient strength, but the thread is more apt to cut through the paper.

Wire

Wire is not much needed by the collector of bird skins, but a small amount of Nos. 11, 16, and 22 should be available for necks of swans, herons, cranes, and other large birds that have long, curved necks. A spool of fine tinned wire is needed for splicing an occasional broken leg or beak of a small bird, and for wiring loose tail feathers.

The mammal collector will need a larger amount of wire of assorted sizes. If "study skins" are to be made up, wire of sizes Nos. 16, 18, 20, 22, 24, and 26 will be needed for wiring the tails. For general use, shrews and small mice require 24 gauge American or English; mice and rats 22 gauge (the size most needed); rats and squirrels 20 gauge; medium-sized squirrels 18 gauge American; large squirrels 16 gauge American or 18 gauge English; rabbit legs, 12 gauge American or 14 gauge English. Mammals needing wire heavier than No. 16 are seldom made up in the field, and only the smallest mammals require a wire as small as No. 26. By far the greatest number of skins will be prepared with Nos. 18, 20, and 22. The ideal wire should be non-corrodible. Galvanized or tinned iron wire, brass wire, and copper wire have been much used. Brass wire and copper wire are generally too flexible to be used in the smaller sizes, and Monel-metal wire has been found to be the best for general purposes. Monel-metal is a bright, whitish alloy (nickel 75 per cent, copper 23·5 per cent, and iron 1·5 per cent) that does not corrode in ordinary liquids (water, brine, formalin, alcohol, etc.). Plain, soft, annealed iron wire may, however, be used without much danger if care is taken to wrap it thoroughly with cotton. An unwrapped iron wire put inside of a moist tail, is almost sure to rust and discolour the skin. For the sake of safety, iron wire may be coated with shellac, which dries quickly, but the wire should be wrapped in any event. Spring wire may be annealed by heating it red-hot and allowing it to cool slowly.

Some judgment must be used as to the size of wire employed, but in general the wire should be heavy enough to give reasonable stiffness to the tail, and small enough to penetrate to the extreme tip of the tail sheath without pulling off the end of the tail. In practice it is often advisable to use a wire slightly larger than the tip of the tail, particularly with long, slender tails like those of jumping mice and pocket mice, and to sharpen the tip of the wire with a file, slightly roughening the tip so that the cotton wrapping will not slip off.

With small mammals, the tail wire is usually the only wire used. If the leg bones are cut off short, or broken, it may be advisable to put a wire on each leg. In rabbits, and similarly long-legged animals, the legs need stiffening and considerable large wire is needed, so that 5 pounds of large wire will not last very long. A pound of No. 22 wire will serve for a hundred or more small mammals.

For large collections of skulls as well as for specimens preserved in liquids it is convenient to have small tags of pure sheet tin or Monel-metal stamped with serial numbers. These tags should be attached to the specimens with Monel-metal wire, which is not affected by water nor corroded by other liquids.

Liquid Preservatives

Alcohol and formalin (formaldehyde) are most commonly used for preserving specimens entire, or for any soft parts, stomach contents, etc., that are desirable to keep. Denatured alcohol or wood alcohol (methyl hydrate) will do for ordinary purposes, but the fumes are disagreeable and often contain acids that tend to decalcify the bones in the course of time. Grain alcohol (ethyl alcohol) is more agreeable to handle and less poisonous, but is more expensive. The alcohol should be at least 85 per cent. If the alcohol is too strong it hardens the tissues, and if it is too weak they decompose, so the better error is on the side of strength. Alcohol or formalin will not readily penetrate thick skins, so that specimens should be cut open on the abdomen before immersion, so that the preserving fluid can get inside. Otherwise a specimen may look good on the surface, but be rotten inside.

All zoological specimens contain a large percentage of water. This is replaced by the alcohol, weakening the solution, so that specimens should not be crowded until they are thoroughly pickled. After the specimens are well preserved, they may be packed more closely, and in some cases the strength of the alcohol may be reduced. If large quantities of alcoholic specimens are to be collected, the collector should have an alcoholometer for testing the strength of the spirits from time to time.

Formalin has some advantages, not the least of which is the light weight. The ordinary commercial solution is about 60 per cent water. For ordinary purposes this is diluted with nine parts of water to make a 4 per cent solution, which may be made either stronger or weaker according to the climate and the nature of the specimen. One great drawback to its use in northern latitudes is that the diluted solution will freeze as easily as water and burst the bottle or containers if they are filled too full. Specimens preserved in either alcohol or formalin become hardened so that they are hard to skin. Formalin also has the disadvantage of contracting the tissues and of bleaching out the colours. Some complaint is made that formalin dissolves the calcium and softens the bones of specimens that are immersed in it for any length of time.

Dr. G. S. Whitby, formerly Director of the Division of Chemistry, National Research Council, Ottawa, was consulted on the subject of neutralized formalin, and the following statement was received:

"If it has been found that neutralized formalin has not the softening effect of ordinary formalin on bones and shells of mollusks it would probably be found more practical and less costly to neutralize the free acid present in commercial formalin rather than ask the formaldehyde manufacturer to do it. In formalin intended for general use the presence of small amounts of formic acid is not objectionable. The practice of adding potassium nitrite to the embalming solution used for preserving cadavers intended for dissection might give good results in the case of formalin solutions for preserving museum specimens. Embalming solutions contain 12-20 per cent formaldehyde, 10 per cent glycerol, and 0·1 per cent potassium nitrite. The object of adding nitrite is probably to oxidize any formic acid present to CO_2 (carbon dioxide) and water. Other methods of neutralizing the acid present might also be tried, such as the addition of calcium carbonate, or sodium phosphate (tri).....I believe there is enough variation in different lots of formalin to make it inadvisable to recommend the addition of a specific weight of the material suggested for trial" (F. G. Green, 1937).

Where the above preservatives are not available, carcasses may be salted or preserved in strong brine. Small specimens may be injected or immersed in a weak solution of carbolic acid or lysol. Specimens in the flesh, or carcasses for dissecting purposes, may also be preserved by immersion in glycerine, or in the glycerine and alcohol solution used by botanists for preserving algae, etc., namely: in proportions, 90 cc. of 70 per cent ethyl alcohol to 10 cc. pure glycerine. Skins may be kept in a soft state for some time by painting the flesh side of the skin with a mixture of carbolic acid and glycerine. In placing animal specimens in any kind of solution be sure that they are first cut open and thoroughly immersed; also that the fur is wet through.

Collecting Cases

A travelling collector who expects to put up a great many skins will find it convenient to have a specially constructed drying box with open spaces on several sides, covered with wire gauze, and fitted inside with several light trays of varying depth. The bottom of each tray is of wire netting, and crossed by movable slats to keep bird skins in place. In such a box skins may be dried while in transit. A smooth-fitting cover of waterproof canvas may be made to slip over the top and sides to keep off rain when moving, and it may be put over the box at night to lessen humidity. This type of collecting case has been used by collectors for the National Museum of Canada for several years (*See* Figure 1).

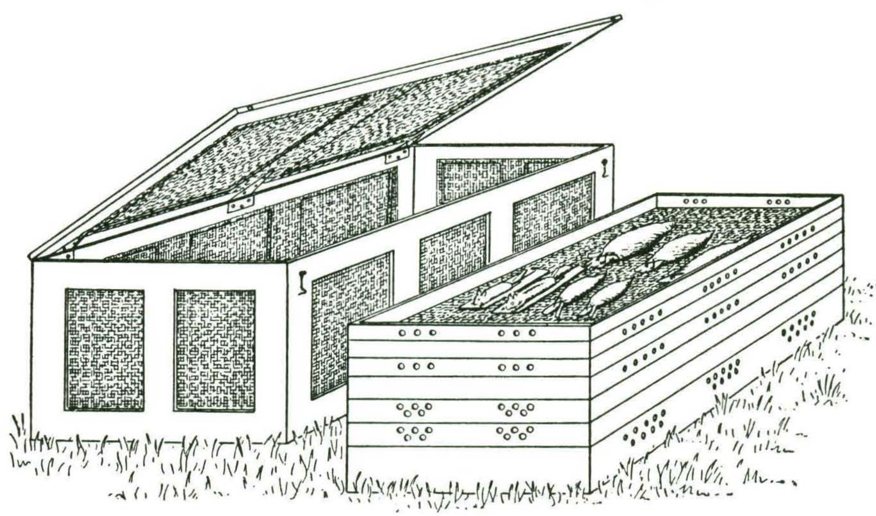

Figure 1. Ventilated collecting case with gauze-bottomed trays.

Gauze-bottom trays may be used in ordinary field-chests, steamer trunks, regulation army locker-trunk, or telescope fibre-cases. When specimens are to be dried, the trays may be taken out and used singly or hung up in stacks by cords at each corner. If trays of greater depth are needed, one tray may be inverted over another one. Blowflies and other insects may be kept out by tying cheesecloth over the trays.

Anthony (1925) describes a type of collecting chest with knock-down trays to be used in a regulation army locker-trunk or steamer trunk. The writer has used this method with some modifications in the standard fibre-cases used by the Geological Survey, Canada, for pack-horse work (Figure 2). The pack-horse case with lower part bevelled off on one side

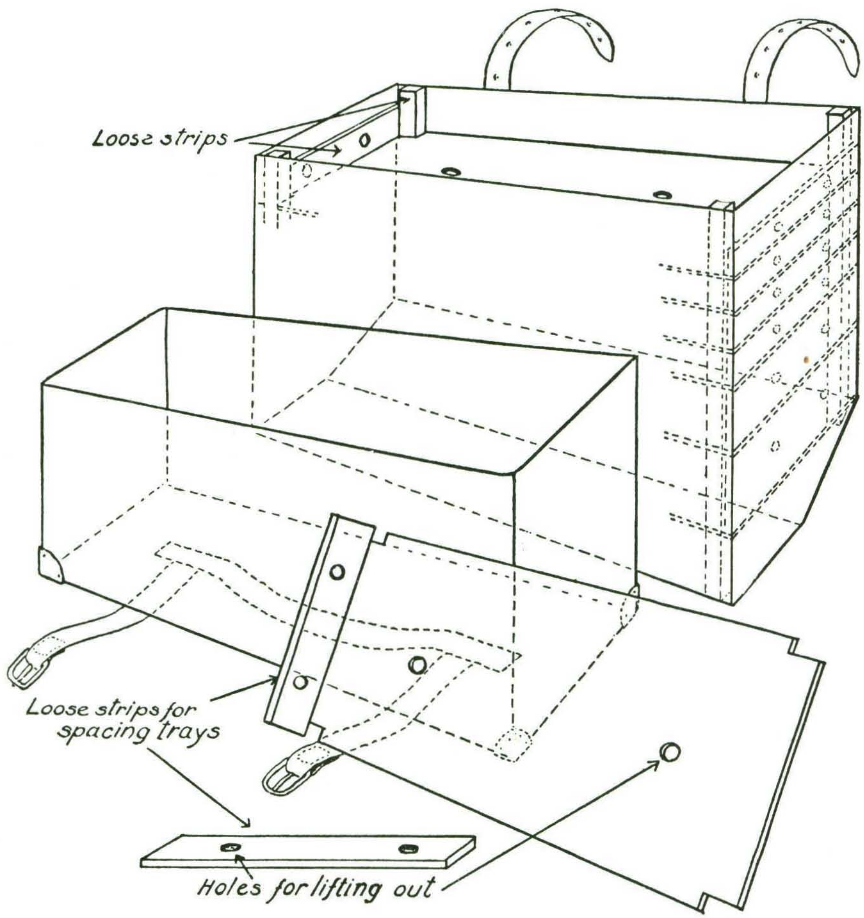

Figure 2. Fibre collecting case for pack-horse work

was found preferable to the old type of rectangular telescope case because it carries with specimens right side up, but any deep, narrow telescope case will answer the purpose. Five or six thin boards $\frac{3}{16}$ to $\frac{1}{4}$ inch thick, of soft pine, beaverboard, or compo-board, are cut so that they will easily drop flat in the bottom of the case. Two strips of $\frac{1}{2}$ inch material as long as the width of the tray bottoms are cut for each board but are not fastened to the board in any way. The width of these strips will govern the depth of the tray and should be such that when the requisite number of boards and strips are assembled, as shown in Figure 2, the last board comes nearly to the top of the case. A useful width of strip for the average small mammal will be $1\frac{1}{2}$ inches, but some strips may be 2 inches, and at least one pair will need to be 3 or $3\frac{1}{2}$ inches wide. The boards and strips, when not in use, lie flat in the bottom of the case, leaving most of the space available for packing equipment. When skins are pinned out on the boards, the boards are spaced one from another by dropping strips across the end. Holes near the ends of the strips and boards allow the fingers to be inserted to lift them out. If the skins are securely pinned into the soft wood, they will not shift during transportation. They should be placed rather close to one another and space (marked off by a line ruled parallel to and $\frac{1}{2}$ inch from the end of the board) allowed for the spacing strip, which will be inserted later. A thin layer of cotton laid upon the skins will make them still more secure when pressed down by the board of the succeeding tray.

Frank A. Bryant, an experienced warden in the service of the National Parks Bureau of Canada, complains that the slippery surface of the standard fibre-case causes difficulty in lashing on pack-horse loads. He prefers a home-made case made of three-ply veneer covered with light canvas, with a strong, tough hardwood cleat screwed diagonally across each end of the case to afford a hold for the pack lashings. Such pack panniers can readily be fitted with light trays or loose pinning boards for specimens if desired. J. Dewey Soper, of the National Parks Bureau, has also devised somewhat similar packing cases and finds them very convenient for carrying tools and specimens in a motor car as well as on a pack-horse.

When a steamer or locker trunk is used, the partitioned tray at the top may be left in and used for tools, supplies, etc., and the tops of the upright cleats cut off to allow for this tray. When using the bevelled horse-pack case, the tools, etc., may be put in the bottom and a supporting cleat cut to fit each end and support the bottom tray horizontally. Cases made in this way are very useful for expendable supplies, such as provisions, as the space can be used later for specimens.

The ordinary light wooden boxes made to hold two 5-gallon gasoline tins are a good standard size for each side of a horse-pack. Specimens may be carried or shipped safely in these by putting in false bottoms made of spare boxes, separated by strips of board nailed to each end. As horses are apt to disjoint the cases by rubbing against trees or rocks, the cases may be made stronger by stitching a stout canvas cover around them. A favourite method of old-time packers was to stretch a fresh calf-skin around the box; this when dried and shrunk in place made a practically indestructible rawhide cover.

CHAPTER II

COLLECTING MAMMALS

SHOOTING

The larger mammals are almost invariably taken by shooting, and some of the smaller species, such as squirrels and rabbits, are more often shot than trapped. Thousands of pages in books of travel and sportsmen's journals have been devoted to the merits of various makes and calibres of rifles, and in general it may be said that high-powered rifles of flat trajectory and soft-point bullets are now almost universally used for big game. The calibre is not so important, as any good sporting rifle will do for most animals in this country. The hunter of lions or tigers is justified in wanting a bullet with a terrific smashing power, but a judicious collector does not want an animal with the skull shattered to bits, or with a great hole blown out of the side. The collector will find a few solid, steel-jacketed bullets useful for shooting smaller specimens, or finishing a wounded animal. The best finishing shot for a large mammal is generally one through the neck vertebræ. If the bullet does not break the neck it will usually sever some of the large blood vessels.

A deer or caribou that is brought down wounded may be cleanly killed as follows. Approach cautiously from one side and grasp the tip of antlers with the left hand. This give a tremendous leverage on the head and prevents injury from a swinging antler. Bend the head sharply downwards. This leaves an unprotected opening in the back of the neck between the atlas and axis (the first and second cervical vertebræ), and by thrusting the tip of a slender-pointed skinning knife through the skin just behind the bony ridge forming the posterior edge of the occipital bone, and down into the spinal cord near the point where it merges into the medulla oblongata of the brain, the animal will be killed instantly. As with many surgical operations, the procedure is simple, but is apt to work more smoothly if preceded by a careful study of the structure of a dead deer. This method is preferable to tearing the animal up with mushroom bullets, or to a slow death by throat-cutting, a method which most sportsmen seem to follow as a tradition of the butcher's trade. If the animal is skinned at once, sufficient bleeding will be done while disembowelling and dismembering, and the animal will be perfectly good for food.

The rifle is not ideal for collecting the smaller mammals as the rifle bullet tears them up too much. A shotgun is indispensable for general collecting of both mammals and birds. A double-barrelled gun is preferable, and shells loaded with different-sized shot, Nos. 10, 6, 4, 2, and BB. A few shells may be carried loaded with a single round ball that will pass freely through choke-bore barrel. This will shoot with fair accuracy for 50 or 60 yards and the writer has killed caribou with ball in 20-gauge shotgun.

When only one weapon is used, the double-barrelled, 20-gauge shotgun is generally the most useful, partly on account of the convenience of reduced charges of shot for small specimens, and above all for less weight (particularly of ammunition) on long trips far from base of supplies. A larger, 16-gauge or 12-gauge, gun is useful on the seacoast, particularly for large water birds. Geese and swans may be killed effectively with the 20-gauge, but the score is not as high, and the missed shots are apt to be forgotten.

An auxiliary barrel, commonly known as an "Aux.", 5 to 8 inches long, and weighing only a few ounces, to shoot ·32 calibre, extra long, centre-fire shell, or ·410 calibre, paper shell, which may be reloaded with No. 10, 12, or "dust" shot, is a great convenience, and saver of ammunition weight.[1] The "Aux." may be carried in the pocket and slipped into the chamber of the gun when needed, or it may be carried in one barrel and a full charge carried in the other barrel. The "Aux." brass shells may be reloaded many times with smokeless powder using nitro primers. If ordinary black powder primers are used, a tiny pinch of black powder should be put in the bottom of the shell before putting in the nitro powder. The wads should be rammed down hard, and the shot wad may be kept in place by a drop or two of melted paraffin wax, or preferably, by a few drops of shellac on top of the wad. Even with the small-calibred "Aux.", the collector will generally find it advisable to load shells with different charges for different animals at different ranges. As with all shotgun loads, it must be borne in mind that at very close range the whole charge will travel as a mass with the impact of a rifle bullet, and it is well to try out the different loads on targets at ranges of from 5 to 30 yards to observe the pattern and penetration and to judge its probable effect on the game.

A good pair of prism binoculars are almost essential to a field naturalist. Mammals and birds may be found at a distance, or identified after observation, and many miles of walking saved in open country. Eight-power magnification is about as high as is practicable in the field, and six-power is high enough for boat work or for use as night-glasses. Higher magnification cuts down both the light and the scope of the field, and twelve-power binoculars are difficult to hold steady. A great degree of steadiness may be obtained by sitting flat on the ground, with both elbows resting on the knees, and steadying the hands and binoculars by resting them on an upright stick or gun case.

Methods of hunting depend almost altogether upon local conditions and the habits of the species wanted. The best times for shooting are early in the morning and late in the afternoon, at the favourite feeding places. Small mammals are generally carried to camp and skinned there but larger mammals may be measured and roughly skinned on the spot, leaving fine work on head and feet until later. If the weather is warm and camp cannot be reached for some time, the body may be cut open and viscera removed. Sometimes it is advisable to inject the body cavity and throat with a weak solution of formalin.

TRAPPING

Most species of small mammals, and some of the larger, must be taken in traps if obtained at all. Most of the fur-bearing mammals are naturally wary or have become shy from persistent trapping, and special measures must be taken to outwit them. Methods vary with the species and local trappers are generally familiar with the details. As professional trappers are usually not very communicative in regard to their "trade

[1] .410 gauge paper shells loaded with No. 12 shot may be obtained from Canadian Industries Limited by special order.

secrets," the collector will often have to work out methods for himself. This is not entirely unfortunate, as in learning to trap, the collector is sure to learn something about the life-habits of the animals sought. This book is not primarily a treatise on trapping nor general natural history, but a few principles may be mentioned for the benefit of the beginner.

For catching the larger mammals appropriate sizes of standard steel traps may be obtained from hardware dealers or trappers' outfitters almost anywhere in Canada. Traps for the larger mammals may be picked for a particular use with much more confidence than would naturally be supposed. The trapper seldom catches a large mammal unless he makes a special "set" for it in the right place. Flying squirrels and weasels often spring marten traps in the woods, and sometimes Canada jays, ravens, and snowy owls will cause trouble. Dogs and cats may also interfere near settlements, but such impediments are not usually serious.

The sizes ordinarily used are No. 0 for catching rats, gophers, and weasels; No. 1 for muskrat, mink, and marten; No. 1½ for fisher, raccoon, and skunks; No. 2 for fox, wildcat, oppossum, etc.; No. 2½ for otter, beaver; No. 3 for lynx, coyote, badger, etc.; No. 4 for wolf, wolverine, etc. Sizes from No. 3 up are generally made with double springs, and smaller sizes with single springs. The larger traps have wider spread of jaws and stiffer springs. A size or two larger or smaller than the ones mentioned are frequently used. A wider jaw spread than usual is necessary for animals with large feet, and if the animal is a strong fighter, like the wolverine, a wide-jawed trap with single spring may not be strong enough. A heavier trap than is needed to hold the animal may injure it unnecessarily and is a dead weight to pack around.

Steel traps are anchored by chains, and sometimes additional strong wire is needed. The ring at the end of the trap-chain may be used for nailing or stapling the trap to a tree, log, or clog, or the trap may be fastened by driving a stake through the ring into the ground. A strong stick may be thrust through the ring and buried in the ground or weighted down with stones, or in the north, buried in a trench in the snow. Snow is tramped down on the stick, and after an hour or so, when the snow crystals have set and frozen together, the stick is held solidly and will have to be chopped loose. A bunch of stout switches will answer as well as a stick and when frozen down will hold a strong wolverine fast. For wolf or bear, where the trap can not be anchored strongly enough to prevent the animal from pulling the trap loose, the trap is fastened to a clog, a piece of log, or stump, which becomes entangled in the brush as the animal drags it along.

Most fur-bearing mammals are taken with a "bait-set", the trap being carefully concealed with leaves, moss, dust, or snow, and the animal is attracted to the vicinity by some kind of bait or scent. Or the trap may be concealed in a path or runway used by the animal, commonly called a "blind-set." If possible the trap should be set so that the catch or "dog" of the bait-pan is on the side opposite to the approaching animal, as otherwise it may step on the catch and be thrown back out of reach of the jaws. In all cases the pan should be adjusted so that it will trip freely, and the trapper should spring it several time to be sure it is going to work properly.

In summer a steel trap may be set in a scooped-out depression, and the space beneath built up with bits of moss, except under the pan of the trap, or a light wad of clean cotton should completely fill the space under the pan, or a thin sheet of paper should cover the trap. The whole is then covered with loose leaves, dust, or any material like the surroundings of the trap. In winter the trap is built up in the same way with moss, dry leaves, or needles of pine, spruce, etc., and soft snow sifted over the trap to make it invisible. A refinement of this method that is found very effective by some wolf trappers in Alaska, who "seal in" the trap scent by sifting a fine layer of dust over the trap before covering it with snow. In very cold weather a firm slab of snow of the proper consistency may be laid over the trap, just clearing the top of the pan, and shaved down to a thin shell. If the snow melts in the day time the pan of the trap may become exposed, or the snow may become softened, freezing later into an icy surface which may prevent an animal from springing the trap. Many trappers carry sheets of thin paper to lay over the trap and sprinkle snow over the whole thing.

"Water-sets" are used for muskrat, beaver, and otter, the trap being set under water. If possible the trap is set in shallows near deep water, and the trap ring held up on a pole set in deep water. When the trap is sprung, the ring slips down the pole, dragging the animal under water to drown. Traps for raccoon are often set under water near the edge of a brook or pond, and a piece of bright tin tied to the pan of the trap is said to be effective in arousing the curiosity of the animal.

Curious animals, like the marten, weasel, and Arctic fox, will often enter little snow-houses or shelters built of branches and twigs, and step into a trap set at the entrance, but most fur-bearers avoid all the works of man. Trapping is a highly specialized occupation, and there is much to learn. Traps need constant attention, as trapped animals will soon spoil in warm weather, and traps freeze up or become drifted over with snow in winter, and must be reset. Dead animals in traps are often damaged by mice, shrews, and large ants in summer, or may be devoured by ravens, jays, or carnivorous mammals at any season of the year.

There are serious objections to the universally used steel trap, principally because large mammals are not often killed at once in the trap and suffer when not quickly removed, but also because many animals get away. However, the steel trap is the chief instrument of the fur trade and the standby of the farmer for catching vermin, and unfortunately no substitute has been devised which is practical and portable enough for the wilderness trapper, and inexpensive enough for the farmer's boy. Steel traps should be cleared as soon as possible for humanitarian reasons. Large mammals will frequently have to be shot before taking them off the trap, and a ·22 rifle will usually do the business. As the scientific collector should at all hazards avoid damaging the skull of his specimen, the best shot is through the neck vertebræ near the base of the skull. Small mammals may be chloroformed, but professional trappers usually kill a fox or smaller mammal by engaging the head with a stick or rifle barrel, so that the animal cannot bite, and then crushing the ribs or neck with the foot. This seems to be rather rough treatment, but causes death as rapidly and painlessly as the deadfall trap or a bullet through the heart, without causing suffusion of blood inside of the skin, as usually happens when a club is used.

Trapping the ordinary small mammals in Canada is a comparatively simple operation. As they are not as a rule pursued by man they are generally not afraid of traps, and the idea is to place the trap in locations where the mammals are found, and where they can see, smell, or step into the trap. The presence of small mammals can usually be detected by tracks, runways, holes, or other signs, and traps should be set out in as many different situations as the region affords. To get a complete collection of the mammal life of any district a trap-line should be run in every life-association area. Every species has a preference for a certain habitat, for purposes of feeding, concealment, or home-building. A patch of grass at the edge of a marsh may appear to be destitute of mammal life, but by stooping and parting the grass with the hands, the naturalist will usually find well-tramped runways littered with "sign" or droppings, bits of cut grass, etc., as plain as a cattle trail, on a small scale. If the trail is fresh a trap set across the runway will usually produce results over night. Traps should be set along streams and ponds for aquatic species, in grass and bushes for meadow-loving forms, at the foot of trees or on logs and branches for arboreal species, and in burrows or at their entrances for subterranean species. The collector may not have the time or sufficient traps to do all this in one day, but the trap-lines may be shifted.

The winter trapper can usually follow his trap-line by his tracks in the snow, but often a fresh fall of snow will hide small traps. At any time the locations of a few traps may be remembered accurately enough, but when many are used the average trapper may have trouble in readily finding them again. Some system should be followed in running the trap-line, as along a path, stream, ravine, or ridge. Trees or bushes may be blazed in a wild country, but in many districts this is not advisable. Small twigs may be bent over, but the writer has generally found it easier to place small cloth markers, or a little bunch of cotton twisted on a bush or stick. The most satisfactory markers are made by tearing white cloth into strips a few inches long and tying these with a loose knot to the nearest bush, or if in a meadow, to a clump of grass. If a trap is set at some distance to one side, and out of sight of the trail, a strip of cloth with one or more knots may be placed by the trail, and a second marker at the trap site. In winter it is better to use markers of bright red cloth as these are easily seen against the snow. Single markers may be used for each trap except the fifth, for which two markers or a coloured mark is set out. This often saves time when searching for a lost trap. When traps are taken up the marker may be placed in the trap jaws for use the next time. If mischievous small boys visit the neighbourhood, it may be necessary to mark the traps less conspicuously.

Many collectors prefer to keep a record of traps set, for data purposes, regardless of their ability to pick up traps by memory. A pocket notebook or a few stiff blank cards of pocket size may be used, and the trap sites noted briefly, something as follows:

1. From bend of creek near camp, 40 paces, runway under log
2. East 20 paces, hole at foot of spruce
3. Back to creek, down 100 paces, on log across creek, etc.

After following such a line once or twice, the notes will probably not need to be referred to except where traps are changed to better locations. The date of running each trap-line should be recorded and it is of interest to record bait used and animals taken at each trap. Such notes on trap-lines will be extremely useful in writing up field notes on mammals, and particularly valuable in checking fluctuations from year to year. Counting up the results of "trap-nights" is the best method known for estimating the comparative numbers of small mammals in the same area from year to year, or the relative abundance at any time. One trap set for one night counts as one "trap-night", and fifty traps set every night for a week, as three hundred and fifty "trap-nights."

The number of traps used will depend entirely upon conditions. Some commercial fur trappers in bush country where the snow does not drift over the traps will use two hundred or more traps and run a trap-line for 100 miles, perhaps taking 2 weeks to make the rounds. On the windswept barren grounds, where traps are either blown clean or buried in hard snow every day or two, the trapper will have hard work to attend to much more than twenty traps efficiently during the short days.

For small mammals the trap-lines will not need to be so long, as the mammals may be at your very door. Moreover, the traps must be visited every day, and in hot weather at least twice daily if possible, otherwise the specimens are apt to decompose, to be polluted by blowflies, or mangled by ants. When ants are very annoying, the writer has found that dusting the traps with pyrethrum insect powder will often keep them away. The powder does not seem to prevent mice and shrews from visiting the traps.

Ordinarily a trapper of small mammals can run a line of fifty or sixty traps, sometimes even a hundred in favourable circumstances. The beginner may often obtain good results by placing small traps at random until, after a little experience, the most favourable places are located. Mammals usually follow natural lines of cover, such as stone and rail fences, fallen logs, or edges of thick bush. Slinking along under shelter, the animal darts across open spaces to the nearest salient of protection. Traps set in such situations may be sprung by the animals in passing even if they are not attracted by the bait. In runways, the trap is preferably set across the runway, and the floor of the runway may be scooped out to bring the trap flush with the surface. Flying squirrels may be caught in traps set on logs, stumps, or horizontal branches, or a little shelf for the trap may be made by placing a slab of bark across dead twigs near the trunk of the tree, or on wooden pegs driven into the bark. The latter method is useful where the lower branches of large trees are high above the ground, as in the western yellow pine. A certain familiarity with mammal habits will soon show what may be expected, as how some mammals will step up to reach an elevated bait, or step over a low obstacle into a trap set on the other side. The solving of exceptions depends upon the keenness or imagination of the collector in understanding animal psychology.

Small traps set in the open should always be tied down. Small mammals are almost invariably quickly killed by shock or by choking, but larger mammals may be caught accidentally and drag the trap away or into a hole. About 2 feet of cord or small copper or brass wire may be fastened to the staple that holds down the trigger, but it is better to drill a small hole through one of the rear corners of the wooden base. Rabbits may be taken with snares of sinew or fine copper wire, and lynx and coyote with heavier wire snares, usually set with a spring pole to jerk the animal up and strangle it.

Several makes of wooden-base traps are on the market and are sold almost everywhere in two sizes, mouse traps and rat traps. The rat size is quite suitable for rats, chipmunks, squirrels, weasels, and the like, but has such strong springs that smaller mammals are often cut in two. The ordinary mouse trap is deadly enough as a simple mouse-killer, but has the disadvantage of being too short and narrow for the scientific collector. A mouse or shrew too often merely has its nose inside when the treadle is touched, and the spring comes down and crushes the skull. As the skull of a scientific specimen is as desirable as the skin, the "Museum Special model trap" (Figure 3) has been devised for scientific collecting. It is

Figure 3. "Museum Special model trap" for small mammals.

intermediate in size between the mouse and rat traps, with width of jaw about 2⅝ inches and spring of about the same strength as that of the ordinary mouse trap. Such a trap usually must be made to order. The "out-o'-sight" and other auto-baited traps depend for their attractiveness on having the wooden treadle saturated with aniseed oil or some other aromatic oil which is very permanent and apparently does not hinder the effectiveness of other baits.

Most of the small traps have wooden bases, and these warp, particularly the cheaper grades, when used on damp ground, or when wet by dew or rain. This can be prevented by dipping the wooden bases into hot paraffin wax or parawax for a minute or two and then draining off. The dipping works better if the trap is fairly warm when dipped, and the wax hot, because then the wax is absorbed by the wood instead of congealing on the surface. Two or three paraffin candles melted in a frying pan will do the work. Paraffin waterproofs the traps and as it is inert is apparently not distasteful to the animals. In very hot desert regions difficulty has been caused by the sun melting the wax out of the wood. Fine sand blowing into this makes a valve-grinding compound which clogs the mechanism and may prevent a small mammal from springing the trap. Shellac, dissolved in alcohol, has been used with success on wooden parts of some of our traps, but extreme care has to be used to keep shellac off the metal parts of the trap as it sets or becomes stiff, and in this way has saved the lives of many small mammals.

The all-metal (galvanized or painted) traps, such as the Schuyler model, are useful, particularly in water-sets, for water shrews and the like, and some makes of small tin "choker" traps are of use for setting against horizontal entrances to holes. Traps which have too much metal surface in contact sometimes cause trouble by "freezing" or cohesion of the parts, and "hang fire" or fail to spring properly. The metal parts of any trap should be gone over occasionally to remove rust and metal bearings should receive a drop of oil occasionally. Although keeping in general to the well-tried standard traps that are in common use, it is well for the collector to experiment a bit and have a few of the odd kinds of traps for special uses. The scientific collector may run across animals that the trap-makers never heard of and is always likely to learn something new while trying to catch them.

One of the principal secrets of success in trapping small mammals is to have plenty of traps and keep them working. The writer has followed a plan of setting small traps in all the likely places, and a few extra ones in unlikely places for the straggler or the unexpected species. Man is far from knowing the habits of all the mammals.

In passing, it may be said that the most effective way to rid houses of rats and mice is to use two or three dozen traps instead of one or two, and set them in every corner, along the baseboards, behind boxes and barrels, and at every hole, so that if the animals miss one trap, they will be apt to find another. With mouse traps at $1 a dozen it does not pay to be sparing of them.

Pocket Gophers

Pocket gophers work under ground and are seldom seen on the surface in the daytime, but their presence in any district is obvious from the piles of small earth pellets thrown up from their workings. The tunnels, which are usually 3 to 8 inches below the surface of the ground, may be found by probing near the mounds, and a section of the roof of the tunnel may be removed and a trap placed in the runway. The hole should then be carefully closed with a slab of sod or piece of board to exclude light. The ordinary steel trap may be used in pocket gopher runways, but much better results can be obtained with some of the special pocket gopher traps that are on the market.

Moles

Moles live under ground in much the same manner as pocket gophers, and their mounds look much alike. Moles feed largely on earthworms and grubs, whereas pocket gophers generally eat roots and other vegetable food. Moles are generally taken in special mole traps, of three principal types: (1) choker loops, (2) gripping or scissor-jaws, and (3) impaling spikes. All depend on the same sort of tripping device, a trigger pan resting on an obstruction produced in the mole's runway where the trap is set. A good strong garden trowel is useful in setting mole traps. The impaling or spearing type of trap is not very satisfactory for collecting purposes, as the skin is usually mangled.

Bats

Bats may often be caught by using a flashlight in caves, mine-shafts, attics, old barns, etc., but sometimes the only method that gives results is snap-shooting on the wing at twilight at the edge of clearings. Mr. C. H. Young informed the writer that he had caught bats in England by suspending fly hooks of smallest minnow size with fine gut leaders where bats are accustomed to fly in the evening. Bats should be collected in all parts of Canada whenever possible, as there is less definite information about the species found, their distribution, and habits, than about the mammals of any other group.

Borell (1937) describes a new method of catching bats by stretching strands of No. 24 and 26 wire across an open water tank, $1\frac{1}{2}$ inches above the water and 4 or 5 feet apart. These knocked down most of the bats that flew over and they were caught as they swam ashore. This method is most practical in a very dry season where the bats come to drink or to catch insects over the water.

Van Tyne (1933) tried out Italian bird-netting methods in tropical jungle in Guatemala, using an imported linen bird net about 40 feet long and over 6 feet high. The two coarse outer nets were made of squares about 7 inches on a side, and the central net of much finer texture had a $\frac{3}{4}$-inch mesh. The whole was suspended by little metal rings from a stout cord stretched between two trees the right distance apart, at about 7 feet from the ground, the space between the trees being cleared. The outer trammels should be taut, and the fine inner mesh then pulled up between the trammels so that it hangs evenly and loosely all over the net. Any bird or bat striking it will make a pocket and become entangled instantly. He considers this method useful in regions where there are no caves or old buildings to harbour bats, or for securing forest bats that do not roost in caves.

Control of Bats

Bats in North America are generally beneficial as destroyers of noxious insects, but certain species often cause annoyance by roosting in attics and summer cottages. The best remedy is to prevent them from entering, by closing all holes and cracks around eaves and chimneys. It is frequently possible to drive bats from attics, double walls, or other enclosed spaces by scattering naphthaline flakes liberally around the place occupied by the bats. Usually 2 to 5 pounds are required to drive the bats from the average roost in a building.

CATCHING ANIMALS ALIVE

It is often desirable to capture small mammals and birds alive, for the purpose of studying poses and photographing them, and to release them uninjured if not wanted for specimens. Sometimes in warm weather a small mammal will spoil in the trap overnight, or a dead animal will be mutilated and destroyed by large ants and other insects that are not apt to injure a living specimen.

The recent world-wide development of bird-banding has brought out so many devices for capturing birds alive, and so many papers have been published on this subject that there is space only to mention Lincoln and Baldwin's "Manual for Bird Banders" (1929), which describes about fifty different kinds of bird traps. Many of these traps are also suitable for capturing mammals, although the ordinary mammal will burrow out of many of the drop traps or ground traps used for birds. For catching mammals the trap will usually need an automatic release, as the small mammals generally go about at night and it is impracticable to watch for them. For rabbits, squirrels, etc., the old-fashioned "Figure-4" trap (Figure 4) is useful. For deadfall purposes the Eskimo fox and

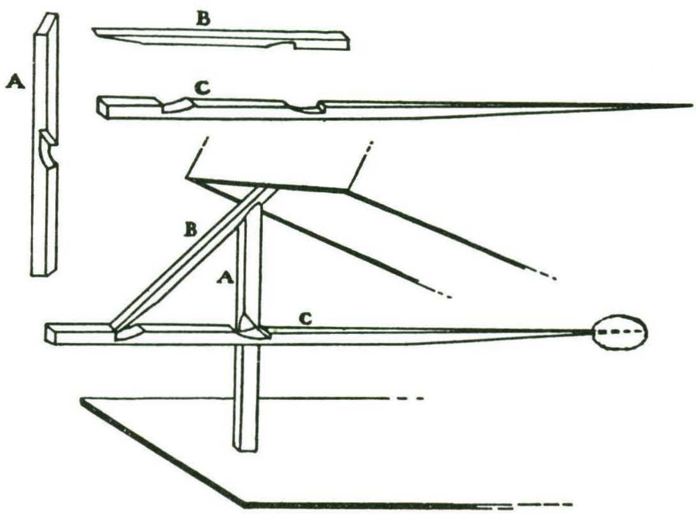

Figure 4. Figure-4 trap.

wolverine trap is still simpler, and might be used to support a drop cage trap. As used by the Eskimo it consists of two short pieces of wood, one horizontal piece with a bait tied to one end, and a shorter upright stick balanced on the outer end of the horizontal stick, with a slab of rock or a heavy log resting on the top of the upright stick. When the animal wiggles the bait the whole thing falls down on him (Figure 5).

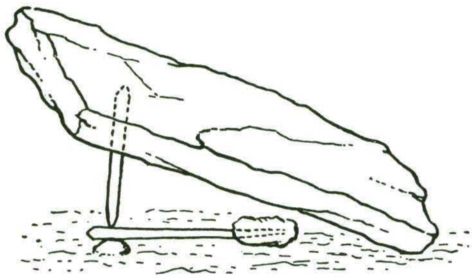

Figure 5. Eskimo deadfall trap.

In many cases the "Biological Survey Cat Trap" may be used to good advantage (Figure 6). It consists of a box about 30 inches long and 12 inches square with a false floor or treadle that rests on a fulcrum (a small piece of wood nailed across the floor of the trap), a trigger wire connected by a loop to a screw-eye at one side of the treadle back of the fulcrum, carried to the top of the trap and passed through a second screw-eye, and a vertical sliding door that is supported, when the trap is set, by the free end of the trigger wire. The door slides in grooves. The

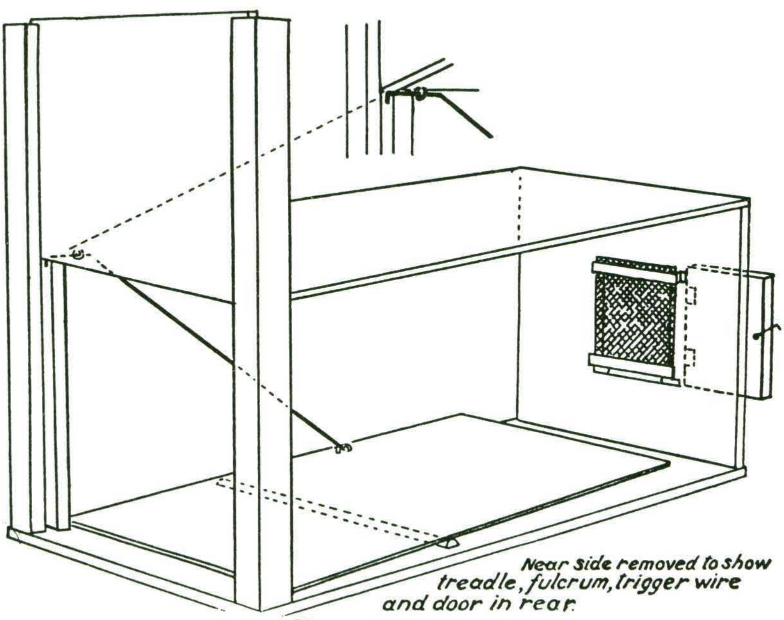

Figure 6. Cat trap.

weight of the cat (or other animal) on the treadle beyond the fulcrum depresses that part of the treadle and pulls back the trigger wire, allowing the door to fall. In the back of the trap an opening about 3 inches square is covered with a heavy wire netting and is provided with a tightly fitting door to permit examination of any captured animal and the introduction of fumigant for its disposal. The door should be open when the trap is set to provide ventilation. Bait (fish is probably the best) is placed well back in the trap. A little catnip will make it more enticing. When the captive is a vagrant cat or other animal that should be destroyed, an ounce of carbon disulphide may be poured onto a wad of cotton batting and inserted into the trap. This will produce fumes that will asphyxiate quickly and humanely. After carbon disulphide has been applied the trap should be kept tightly closed to confine the gas. Carbon disulphide is highly inflammable and explosive, and its fumes are offensive and poisonous if inhaled in a closed place. It is, therefore, advisable to use it only in the open air. The use of chloroform as a killing agent is preferred by many persons, and it may be passed down into the trap through a tube from a scent spray.

The various fur-breeders' journals and fur-farmers' outfitters advertise numerous traps for catching beavers, muskrats, mink, marten, skunks, and other fur-bearing mammals alive for breeding purposes, but the general collector will not as a rule care to keep an assortment of all the new specialties in this line (*See* Ashbrook, "Fur-Farming for Profit," 1928).

Vernon Bailey (1921-1932) describes some simple devices for catching small mammals alive which will be useful for any field collector. Mr. Bailey stated that with a pair of pliers and a small coil of spring wire he could go to any garbage heap where there were plenty of tin cans and make as many mouse traps as he needed in an hour or two.

The simplest trap is an inverted bowl, tin can, pan, bucket, or box. A light bowl may need a stone on top for weight. A trigger, rounded at one end and pointed at the other, is cut out of a thin board or shingle. Fasten some bait to the pointed end and place the rounded end under the edge of the bowl and the baited end about the middle underneath. The mouse wiggles the bait and the bowl drops over him. A paper, tin, or bag may be slid under the bowl and the mouse picked up. A tin can makes a good live trap. Cut a piece of tin to fit the inside of the open end. Hinge it with a wire loop at one edge so it can swing in and not out. Fasten a springy wire to the lid and to the outside of the can so as to act as a spring that will pull the lid shut. Push the lid in and place a baited spindle under its lower edge to hold it open until the mouse wiggles the bait. Then the spindle drops and the door snaps shut with the mouse inside. A still simpler trap may be made by cutting a hole in one end of a tin box and placing a sloping hinged door inside that easily lifts up for the mouse to enter and drops down behind him.

Some small mammals are easily caught by sinking a deep tin can, jar, or bucket in the ground along a runway, leaving the top just level with the surface of the ground.

If a garbage hole is dug for camp slops it is well to look into it now and then for small mammals that may come to investigate a new source of food supply. Mr. Stuart Criddle, of Treesbank, Manitoba, writes that he frequently uses pitfalls for catching mice, making them round, about 14 inches across at the surface, 24 to 30 inches deep, and larger at the bottom than the top, so that the mice have no chance of climbing out; also that sunken tins or glass jars filled half full of water give good results in dry weather, and when placed in muskegs where the water is close to the surface. He has also found that holes made with an 8-inch post-hole digger are satisfactory. The earth from the pit should be placed on either side of it. The mice will go to play on the sand piles or pass between them and fall in. Grain or oatmeal may be scattered about the pits, but this is not required if the pits are placed in the right place. If the animals are wanted alive it is well to place a tin can of meat and other food in the pits. Pits should always be visited the last thing at night and as early as possible in the morning. Mice may also be caught alive by tying a sheet of paper over a wide-mouthed jar or pail, and cutting two long slits in the paper, crossing at the middle. Crumbs or other bait are placed on top of the paper and the weight of the animal causes it to fall through. C. E. Johnson has seen a variation of this method used in Quebec, for catching rabbits. A keg or barrel with lid on a swivel hinge in the middle is buried in a snowdrift and a piece of cabbage suspended over the lid. He also notes a method of catching muskrats used at St. Thomas, Ontario, in which a keg with a small amount of water and some apples in the bottom is sunk with rocks until the top is near the water-level.

Clarke (1938) describes a method of using "water-traps" that is inexpensive, and for some species, particularly shrews, jumping mice, etc., was strikingly more effective than the use of ordinary mouse traps. The water-trap is made by sinking a bucket, tub, or large tin in a hole dug in ground deeply enough to have the rim of the vessel below ground level. If the ground is high and dry the bucket must be tight and have its own water supply. In low sites holes may be punched in the bucket and water allowed to seep in. The advantages of the water-trap may be listed: (1) means of collecting small mammals with a different selectivity from mouse traps; (2) trapping may be repeated, except in winter, under identical conditions; (3) no bait is required; (4) once established, a set runs itself; (5) if the weather is cool the hair of specimens is kept from slipping for some time; (6) no specimens damaged and no traps robbed; would-be robbers are themselves taken; (7) they may be used for moles and in other runways. The main disadvantages are in being cumbersome, and that the collector of ectoparasites can not be sure of his host records. Water-traps were more efficient in certain situations, such as along streams. Dr. Clarke suggests that water-traps could be made in portable units, with the traps nested, a bottom on the outermost, and then every second or third trap bottomless. Those lacking bottoms could be set where there is a natural water supply. The depth of metal in the trap need not exceed 1 foot.

POISONING MAMMALS

The use of poisons is not recommended for killing mammals for specimens. The animal often wanders away after taking the bait, and many are never found, or if found are usually spoiled. There is also danger of killing other harmless and useful mammals and birds. The only justifiable use of poisons is in control of injurious rodents such as ground squirrels, pocket gophers, etc., that are damaging crops, and possibly in some cases for certain species of predatory mammals. At the proper season and by exercising care poison may be used on infested farming areas without doing much damage to other forms of life, but the possibility of destroying domestic animals and valuable wild fur-bearers must be taken into consideration. In Canada the use of poisons for such purposes is strictly regulated by Federal, Provincial, and Municipal laws and regulations.

BAIT

For the carnivores, where blind sets are not used, the best bait is the favourite food of the animal, although where prey is abundant the bait may not have much attraction. For winter trapping the Eskimo trick of freezing a fat ground squirrel in autumn and employing it on the trap-line is useful. After the trap is set a few, tiny frozen chips are hacked off with the hunting knife and scattered carelessly on the snow. These bits have a pungent, ratty odour, and cause the fox to look for more and in the search he may step on the trap. One squirrel or gopher will serve for hundreds of sets. Most professional trappers have various malodorous recipes for attracting marten, fisher, mink, and the like, depending on mixtures of beaver castoreum (or "castors"), decomposed fish, fish oil, rotten eggs, and the like. Dead carcasses will also attract many of the carnivores.

Young (1930) recommends a scent for attracting wolves and coyotes, made as follows:

"Put into a bottle the urine and the gall of a wolf or a coyote, depending on which is to be trapped, and also the anal glands, which are situated under the skin on either side of the vent and resemble small pieces of bluish fat. If these glands can not be readily found, the whole anal parts may be used. To every 3 ounces of the mixture add 1 ounce of glycerine, to give it body and to prevent too rapid evaporation, and 1 grain of corrosive sublimate to keep it from spoiling. A few drops of the mixture should be scattered on weeds or ground 6 or 8 inches from the place where the trap is set. A little of the scent should be rubbed on the trapper's gloves and shoe soles to conceal the human odour."

If the animals become "wise" to this scent an effective fish scent may be prepared by grinding the flesh of sturgeon, eel, trout, sucker, carp, or other oily fish in a sausage mill, and leaving in a warm place of even temperature to decompose thoroughly. This scent may be used within 3 days after it is prepared, but it is more lasting and penetrating after a lapse of 30 days.

For lynx and bobcats, Young (1931) recommends scenting with the fish bait just described, but several modifications have been found highly effective. To the decomposed fish as a basis may be added mice, beaver castors, musk glands from minks, weasels, and muskrats, and the bladders of coyotes and bobcats. Oil gives body to the scent and to a certain extent prevents freezing. If the mixture appears to be too thin, glycerine, brains, fish oil, butterfat, or other animal fat, such as that from woodchucks and ground squirrels, may be added.

Oil of catnip, diluted in the proportion of 36 drops of the pure oil to 2 ounces of petrolatum, has proved an effective lure for bobcats as well as lynx and mountain lions. If the pure oil is not obtainable, catnip leaves boiled to a pulpy consistency in water may be used.

Arthur ("Fur Animals of Louisiana," 1928) gives a list of baits used by trappers in Louisiana, and some of these should be useful to our collectors who wish to collect material in the summer time:

"Muskrat—Muskrat musk mixed with anise oil and oil of rhodium.
Mink—Best is musk of mink; also fish oil, or mixture of mink and fish oil.
Skunk—Tainted meat, especially tainted skunk and rabbit.

Raccoon—Fish oil mixed with a few drops of anise oil and a little honey. Also fish oil, muskrat musk, anise oil.

Weasel—Fish oil mixed with anise oil, asafœtida and oil of rhodium.

Otter—Fish oil, mixed with anise oil.

Opossum—Food, bird carcasses, rabbit meat or carrion.

Fish oil prepared by cutting fish into small pieces and placed in uncovered jar in the sun to decay. The oil should be poured off and corked in small containers.

Scent should never be placed on the pan of the trap, but should be placed on a stick above the trap, being arranged so that the animal will have to step on the trap to get the scented bait."

Skunks may be caught in traps set above an animal carcass buried just below the surface of the ground, or in a trap set at the foot of a tree with a carcass suspended a foot or two above the ground. A tainted trap in which a mink has recently been caught is apt to attract other minks in the neighbourhood.

Most collectors have found pikas (*Ochotona*) difficult to trap, but Mr. Kenneth Racey of Vancouver showed the writer several beautiful specimens in winter coat that he trapped at entrances to holes in snowdrifts above their talus slope retreats. He states that the pikas sometimes come out in early winter and at times travel 50 feet or more to a bare rock exposed above the snow. Mr. Walter W. Dalquest (1939) states that he has had good success in trapping pikas at edge of talus slides, by using dried prunes for bait in rat traps.

Some authorities advise baiting all small mammals with their natural food, but their food is not always known, and in the proper season the natural food is generally so plentiful that the animal will not have to go into traps for it. The same applies to most vegetable and fruit baits.

Rolled oats are a standard bait for most rodents, although the writer has never found lemmings or tundra voles much attracted by it, and no wild animal in his experience seems to like beans, oatmeal porridge, or oatmeal when soaked by rain or dew. Loose bait of this kind should be sprinkled sparingly over the treadle and in front of the trap. Dried fruit, bits of vegetables, bread, etc., are taken by some species. Scraps of bacon rind, cheese (the older the better), and dried fish are attractive to shrews, and are also liked by mice. Flying squirrels like pieces of fresh apple preferably cut so that the core and seeds show. Fresh fish, fresh meat, or carcasses from the skinning table may be used as bait for shrews and carnivores.

The writer is accustomed to carry three or four small boxes of combination baits when trapping small mammals, one containing a mixture of rolled oats, bread and cake crumbs, and a few aniseeds, or a few drops of oil of aniseed or oil of caraway. This is a good bait for any kind of mice or voles. Another box is used to cater to shrews, and contains little pieces of raw or fried bacon rinds, bits of cheese, peanut butter, and dry fish; and if possible another box of bait the same as the last but flavoured by a few drops of oil of valerian or tincture of valerian. The valerian odour is attractive to insectivores, small carnivores, and apparently to mice also; at least mice are not repelled from the traps by the smell.

One of the best all-round baits consists of a mixture of rolled oats, chopped-up raisins, and oily peanut butter; it has proved successful in temperate regions and in the tropics. Anthony (1925) gives as his most successful combination bait: "One part bacon, cut up into small pieces; one part of cluster raisins, also cut up small; two parts of oily peanut butter; rolled oats sufficient to make the mixture of putty-like consistency." If thoroughly mixed the bait will keep for years in a tight jar or tin. This combination bait is attractive to rodents, shrews, opossums, etc., and because of its oily nature will not be washed away by rains. These soft mixed baits are usually pressed into the hole in the bait treadle. Some baits, such as pieces of meat, fish, or fruit, may need to be tied to the treadle with fine thread or wire. They should be tied well back, so that the trap will not be sprung too soon.

LABELLING SPECIMENS

Specimens should always be fully labelled at the time they are prepared, as a specimen without an authentic pedigree has very little scientific value. It has been said that a good label without a specimen has a certain amount of value, but a specimen without a label has almost none. As the original label should always be preserved in connection with specimens, no matter how many subsequent additions are made, it should be of moderate size, never exceeding 3 inches in length and $\frac{3}{4}$ inch in width, and neatly and legibly written. If possible, use the permanent label of the museum or other institution for which the collection is being made. If ink is used for labelling, it should be India ink or waterproof carbon ink (such as Higgin's Waterproof Ink or Higgin's Eternal Ink). Ordinary writing or fountain-pen ink or ball point pens should never be used, as labels are very apt to become damp or grease-stained; then the ink spreads and the writing becomes illegible. The lettering should be done with a fine pen, and preferably in print. If waterproof ink is not available, the labels should be written with medium hard black pencil (never "indelible" or "copying" pencil), and the legend may be later traced in ink. When writing on either side of the label, always keep the string to the left; this facilitates its reading (*See* Figure 19).

If ordinary price-tags are used as labels for skins, care should be used to obtain tags with white strings. Many specimens have become badly damaged by coloured strings becoming wet and indelibly staining the hair or feathers. Heavy manila shipping tags are very good for large, heavy specimens. These can bear the full data, but as insurance against the tag becoming torn off in shipment, it is well to attach an additional, numbered tag of metal, leather, or wood, or to mark the inside of a dry skin with soft pencil or India ink. Beware of cloth tags or handle them with caution. The writer had the experience of hanging several mountain sheep skins, bearing cloth labels, up to dry and of finding that the rain and melting snow washed the glossy sizing out of the labels and the legends with it. Labels or tags should be firmly tied with a double knot. A half-hitch, slip-knot, or granny-knot is not to be depended on.

The data of small skulls may be written on labels of strong, tough paper, with heavy black pencil. The collector's field number (corresponding to the number of the skin) and the collector's initials are usually enough if the skin accompanies the skull. The number should be written on both sides of the tag, in case one side becomes obliterated, as imperfectly cleaned skulls may become soft in damp weather. If the skull is alone, it may be well to put the usual complete data on the accompanying label. Some collectors have a careless habit of putting nothing but their catalogue numbers on the skins; but field notebooks are frequently lost and then it is almost impossible to identify the specimens accurately. The initials are useful in case several collectors are sending in material. For large collections of skulls it is convenient to have small tags of sheet tin or Monel-metal stamped with serial numbers. These may be attached to the skulls with Monel-metal wire, and are not affected by water or corroded by other liquids. For large skulls and bones a piece of sole leather or any heavy leather makes the best labels and the number and initials may be carved with a jack-knife. A smooth piece of wood also makes a very good label and any desired data may be carved on it. The only caution is not to use too thin a piece of wood, which is liable to split and come loose from its fastenings. The strength and durability of a label should, of course, be in keeping with the stress that the specimen has to undergo. A mouse or bat is small and light, and will not stand much strain without destruction and must be properly packed, whereas buffalo, walrus, and whales will probably have to undergo heavy handling before reaching the safe haven of a museum.

Although the field notes of a good collector, and sometimes the "remarks" in the field catalogue, will contain notes on the character of the area in which the animal was taken, that is, "sparsely wooded hillside," "sphagnum bog," "salt marsh," etc., in addition to approximate altitude, Hamilton (1938, page 102) urges that as much detail as space allows should be recorded on the specimen label where it is more apt to be available in the future. He suggests noting with *sex*, reproduction condition; *stomach contents*, if not saved, note what can be determined by cursory examination; food contents of *cheek pouches;* and *parasites,* the presence or absence of fleas, mites, ticks, roundworms, bladderworms, and their relative abundance. The study of animal parasites is becoming of great importance in both human and veterinary pathology, and specimens taken from definitely known hosts should be preserved when possible, and noted on the "host" label.

Every separate part of a specimen—skin, skull, loose bones, or sections of a skeleton—should bear the same field number, and at least one part, preferably the skin, should have a label firmly attached, carrying the following data, of which items Nos. (2), (3), and (4) are absolutely essential.

(1) Collector's field number.
(2) Locality, or place of capture.
(3) Date of capture.
(4) Sex of the specimen.
(5) Measurements of the specimen.
(6) Collector's name.
(7) Colours of soft parts; eyes, lips, and any areas not covered by hair.

FIELD CATALOGUE

A catalogue or notebook containing a list of specimens collected should be kept and each specimen numbered as it is prepared. This number should be written in the catalogue, on the label tied to the skin, and on the label fastened to the skull. The catalogue or the field notes should contain any pertinent data for which there is not room on the label. To avoid omitting any data, it is well to rule the leaves of a good stout notebook before going to the field. The writer's plan is to leave four lines across two opened pages for each specimen. On the left hand page rule vertical columns for: (1) Collector's field number; (2) Locality; (3) Altitude; and (4) Date. On the right hand page rule vertical columns for: (5) Sex; (6) Name of species and common name, colour notes, habitat, etc., and (7) Measurements.

The locality should be so indicated as to be intelligible to anyone with ordinary geographical knowledge, and should be stated with reference to some definite, well-established geographical locality, province, territory, or district. In settled regions the county and the location in respect to nearest post office will do, but in unorganized territory, the locality should be connected with some officially named river, lake, or mountain, or, failing these, the latitude and longitude may be given. Names like "Goose lake," and "Bear creek," are so common that they mean little by themselves, and names like "Smith's ranch" are transitory. If the region has high relief, altitudes should be given if possible, as different forms of life are found at different levels and the elevation may give the clue to the life zones. Altitudes may be obtained from maps, or in some cases from railway time-tables, and local heights may be estimated from the nearest known elevation.

In writing dates, spell out the name or abbreviation of the months and do not express them by numbers. A common but reprehensible practice is to write month-day-year, or day-month-year, in figures, but there is no uniformity about the custom, and one is left in doubt whether 3-2-32 means March 2 or February 3. As it is confidently expected that many of the specimens are permanent enough to be extant more than 100 years from now, the year had also better be written in full.

DETERMINING SEX OF MAMMALS

The importance of carefully and unmistakably determining the sex of every mammal specimen and of marking the sex on the label can not be too strongly emphasized. In making scientific studies it is essential to be sure of the sex of specimens examined, as the presence or absence of primary and secondary sexual characters must be taken into account. Males and females frequently differ in colour and may moult or shed the coat at different seasons. The sexes usually show differences in dimensions of skull and bones, in total length of body, length of feet, and the differences often grow more pronounced as the animals grow older. In some of the less well-known species, these differences have not been well worked out, and it is important to have numbers of accurately sexed specimens to determine these differences.

Some of the variations from the ordinary type of mammalian sex organs have been described in works on zoology and comparative anatomy, and an experienced anatomist can work out the exceptional cases by the light of reason. Unfortunately, the average field collector is not always deeply versed in anatomy, does not usually have the books at hand, and frequently does not know the name of the species which he may be working on. Mammal collectors of many years' experience have been found groping in darkness while attempting to determine the sex of small mice or shrews and even of larger mammals.

The sex of large mammals and most of the smaller ones can almost always be easily recognized by the external organs of animals in the flesh. With the larger mammals the sexual characters may frequently be noted on mutilated dried skins, or by the development of the skull, horns, or other appendages, but on account of warping and shrinkage of skins in drying or tanning, conditions are often distorted and difficult to observe with any degree of confidence, so the sex should always be determined from the fresh specimen and recorded.

In small mammals the sexual characters are often small and obscure, particularly in young specimens in which the organs are undeveloped. In the first place, an animal which is cleanly killed, without blood clots, bruises, or discoloured organs is easier to examine. Torn viscera, intestinal juices, and decomposition will add to the other difficulties. If the animal is allowed to remain for some time, depending on the season, until the rigor mortis (stiffness of freshly killed animals) has passed off, there is not so much bother about blood flowing and obscuring the organs.

Many of the smaller mammals have the sexual organs in both sexes greatly enlarged during the breeding season, and at such times the testes of adult males are generally conspicuous from the outside, and in cases of doubt may be easily verified by dissection. The nipples of nursing females are generally noticeably enlarged, and the fatty, whitish coloured mammary glands are conspicuous and diagnostic when the skin is removed. The nipples alone are not a perfect guide to the sex as they are frequently found in a vestigial state in the male of the species.

In some mammals, particularly mice and shrews, during the intervals between the breeding seasons, and in young specimens, the sexual organs are small, shrunken, or undeveloped, and the external genitalia are superficially much alike in the sexes. The male sheath, or tubular fold of skin into which the penis is retracted, looks much like the external orifice of the vagina. The organs are obscured by hairs, and frequently these parts are so close to the anal aperture that the structure cannot be differentiated without use of a magnifying glass. In such cases, the fold of skin should be picked up with fine-pointed forceps and manipulated or forced back, and in the case of a male the organ will protrude, looking like a small white thread. In some cases ossification may be detected, but the organ is so slender that the condition is hard to observe in gross dissection. If still in doubt, the presence or absence of the testes should be verified as the animal is being skinned. The testes are whitish or yellowish organs when not discoloured by ingested blood, and in a shrew may not be larger than a grain of sand. Care should be taken not to confuse the testes with grains of cornmeal or sawdust used in the skinning operations. The use of a small hand lens or magnifying glass is necessary in some cases, and should be a part of every collector's and naturalist's equipment.

The condition of the reproductive organs—state of testes, whether small, slightly enlarged, or greatly enlarged—may be shown by an outline sketch drawn on back of the label; whether testes are descended or not descended; condition of the mammæ (lactating); presence or absence of embryos. If embryos are present, their number and size should be indicated.

The smaller mammals frequently have several litters of young during the warm season in the North, or at any time of the year in more southern regions, and their fertility is of great interest in connection with climatic conditions, food supply, and other variable factors. The size of the fœtuses will give some idea of the relative date of birth.

In the beaver and the porcupine, the testes are inside the body skin and hidden by thick folds of fatty tissue. Fur farmers and others often wish to determine the sex of captured living animals, and it is well to know that even if the male organs are not noticeable from the outside, the testes may usually be detected by pinching hard and deep with the fingers. By pressing down hard on the abdomen in the region of the sheath, the male organ may be forced out and easily recognized. In skinning dead mammals of this size, the organs are, of course, readily picked out (*See* Young, 1936).

Never put the sex on the label unless certain of the fact. A question mark (?) after the sex mark should indicate any doubt on the subject, and even experienced collectors have to do this at times. It is a scientific crime to put down inferences as facts. The symbol ♂ (the astronomical sign for the planet Mars) is generally used in zoology and botany to mean male, and ♀ (the astronomical sign for planet Venus) for female.

If the age of the specimen is known, indicate it on the label. If adult, as shown by bones, skull, horns, or coat, it should be marked "adult" (or ad.), or if young, "juvenile" (juv. jv.). The age is often known by observing young animals with their parents.

The measurements of specimens are of considerable importance as most of them must be taken in the flesh and cannot be obtained from a dried skin. As most modern scientific works on mammals, in this country as well as in Europe, give measurements in the metric system, measurements, particularly of the smaller mammals, should be taken in millimetres whenever possible. It is much more convenient to compare lengths in millimetres than in inches and minute fractions of an inch. Rules graduated to both inches and millimetres are easily obtained. A convenient way to reduce inches to millimetres, or vice versa, is to fit a stiff paper collar around a steel inch-millimetre rule, when by sliding the collar to any fixed point on the scale the corresponding equivalent in the other system may be read off without calculations. Where many measurements have to be correlated, it is convenient to have on hand a table giving corresponding equivalents for inches and millimetres.

MEASUREMENTS OF MAMMALS

For general information and for purposes of scientific comparison with other specimens, *three measurements should be taken of every mammal, namely*:

(1) *Total length* (abbreviated as L.)—the distance in a *straight line* from the tip of the nose to the end of the last tail vertebra, exclusive of the hairs. If rigor mortis (the stiffness which sets in shortly after death) has contracted the muscles, the body must be stretched and the limbs pulled into a natural position. Place the body on its back, hold the tip of nose at the edge of board or table, or against a pin, or anchor it by a pin through the nose, hold the body down firmly, pull the hind legs out firmly to full length, and set a pin to mark the end of the last bone of the tail. The distance between these two points should be measured with rule or

tape and set down at once on the label or in the notebook (preferably in both places, as a label often becomes detached in process of drying specimens). A wooden rule, marked in millimetres, with a small brass or tin plate screwed at right angles to one end will be found convenient for measuring small mammals. The nose may be shoved against the end of the rule, the end of the tail located by thumb or finger nail, and the reading taken directly from the ruler without the aid of pins (Figure 7). Very often a small mammal will have the back broken by the trap, and the two sections of the body hanging limply, connected by the skin. By shoving the parts together an approximate length may be measured, but the conditions should be noted on the label.

(2) *Length of tail* (abbreviated as T.)—the distance from base of tail to tip of the last vertebra, exclusive of the hairs. The tail may be bent at right angles to the back, and the length taken with a stiff ruler or a pair of dividers. A convenient method is to hang the body over the edge of a table, with the tail flattened out on the table, and mark the distance with a pin. With mammals like the pika (*Ochotona*), on which the tail is so short as to be hardly noticeable through the skin, or in cases in which the root of the tail is very thick, inflexible, and hard to locate, merging imperceptibly into the body, as in the porcupine, the tail measurement is best taken from the skinned carcass, and the fact noted on the label.

If the length of the body is desired, it may be obtained by subtracting the length of tail from the total length.

(3) *Length of hind foot* (abbreviated as H.F.)—the distance from the end of heel bone (calcaneum) to the end of claw on the longest toe. This is sometimes recorded as "length of hind foot, *cum unguis*", or "length

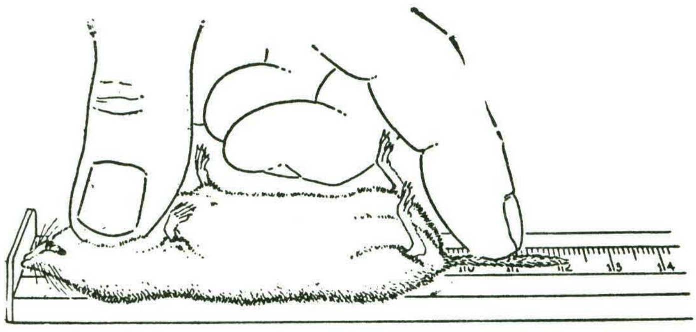

Figure 7. Measuring total length of a small mammal.

of hind foot, c.u.", to distinguish it from the usual European measurement of only to the fleshy tip of the longest toe, i.e., "length of hind foot, *sine unguis,*" or "length of hind foot, s.u.". The toes should be straightened out and this may be done by pressing them flat against the ruler, or the foot may be pressed flat on the table and the length measured with dividers.

The length of the hind foot is in many ways the most useful measurement of all, as this length is subject to less individual variation in specimens of the same species and age than any other superficial measurement that can be made, and, what is of practical importance, the measurement is fairly uniform as recorded by different collectors. This is an important consideration in small mammals in which a difference of 2 or 3 millimetres in length of foot is an important specific character, being really 10 per cent when the normal is 20 or 30 millimetres. With the larger mammals, the same collector may get fairly uniform results in measuring lengths of body and tail, but other collectors may have a different technique and get widely varying results when measuring large mammals on uneven ground, or when the specimens are distorted and shrunk by rigor mortis. For this reason the foot measurement should not be neglected.

Another method of measuring is to prick off each measurement with dividers on board or paper table cover, measure them with steel tape or rule, and enter the figures on label. Whatever method is adopted, it is well to acquire a regular habit in this necessary routine, so that measurements may be uniform and no omissions made. The three essential measurements (length, tail, and hind foot) should always be recorded in the same order, as for example: "L. 221; T. 129; H.F. 31," usually shortened on the label to "221-129-31," the use of millimetres being assumed.

The height of the ear above the crown of the head often affords a valuable measurement, particularly in bats or long-eared mammals, and is written on the label, "height of ear above crown." It is taken from the crown of the head at the base of the ear, to the tip of the ear, exclusive of any tufts of hair. Some collectors take measurement "ear from notch," that is from the notch at the lower opening of the ear conch to the tip of the ear not including hairs. Bats should also have "height of tragus" (the flattened eminence of the auricular front of the opening of the external ear). It is a good thing to make a pencil sketch of the outline of a bat's ear, on the back of the label, as the ear is apt to shrivel up in drying if not carefully watched.

If the mammal is to be mounted, particularly if it is of large size, a number of other measurements should be recorded for the benefit of the taxidermist; details of these measurements are given under heading of "large mammals" (page 65). When small mammals are taken, to be mounted later, it is well to preserve the skinned body in alcohol, formalin, or brine, or failing these, to make tracings of the body and limbs.

The weight of animals, alive or "in the flesh," is of considerable interest and in many cases controversial because of the neglect of the subject by most collectors. If possible, weigh small mammals in grammes with balance scales. Medium-sized mammals may be weighed to ounces with a tested spring-balance or milk scale. In spite of generations of big game hunters, the amount of authentic data in regard to the actual weight of many of our larger mammals is surprising, for as Seton (1929) points out, it is difficult to get the scales and the animals together, and hunters' guesses are of the same order as fishermen's. Any collector who has the opportunity is urged to add to our information on verified weights of large mammals. It is desirable to weigh the body as a whole, but where this is not practicable to weigh the animals cut up piecemeal. In weighing animals after cutting up, try to weigh the entrails as well as the flesh, and make an estimate of weight of blood lost.

The collector's name should appear on the label as authority for the facts stated, and in order that due credit may be given. It is often important to know the collector's name in case any further information is desired about the specimen.

The collector will often find it desirable to provisionally add the name of the species (as bat, mouse, shrew, etc.) for his own convenience in listing or referring to the specimen in his notes, but exact specific or subspecific determinations are not necessary or desirable because field identifications are very apt to be wrong, and every species has characteristic marks by which it may be known by the experts. If the species has a local name by which it is known to settlers or natives of the country, it is urged that this name be recorded on the label as well as in the collector's notebook.

CHAPTER III

SKINNING MAMMALS

SMALL MAMMALS

Labelling

The first thing to do is to prepare a label or at least make notes of certain data which can only be obtained from the fresh specimen "in the flesh." Locality, date, and sex, should be put down first of all; then the three essential field measurements: length (L.), tail (T.), and hind foot (H.F.), taken in millimetres if possible. Take the measurements in inches and fractions if a millimetre rule or tape is not at hand. Colour of eyes, and of any soft or hairless parts that are apt to change colour on drying, should be noted on the back of the label.

Cased Skins

The simplest way to prepare small mammal skins is by "casing" them and this method may be used for any mammal from the size of a shrew up to the size of a wolf. The "cased skin" is easily and quickly prepared, without the use of preservatives, and with no other tool than a knife, and is quite suitable for scientific purposes. The student of seasonal moults and colour will often find skins "cased" with the flesh side out useful in showing the localized areas of new hair growth that form variegated patterns which are well known to fur buyers as evidence of "unprimeness" of the skin, but which are often hardly visible on the surface of the fur (Figure 8). The "cased skin" is recommended for the casual collector or traveller who wishes to preserve an occasional specimen, or a collector who is pressed for time, or who is travelling light. In briefest terms:

Skin the animal in the same way as a trapper would skin a fox or mink, leaving claws and feet attached to the skin, and dry and preserve the skull separately.

One opening cut is made, beginning at one heel, cutting through skin at back and inner side of leg, across base of tail between the anus and the urethral opening, and down to opposite heel. Detach skin from legs, cut through each leg a little above heel, peel skin down on feet as far as the toes if possible and cut away any loose flesh, and loosen skin around base of tail. The skin of the base of tail is seized with the thumb and finger of one hand and the tail which is attached to the body is pulled with the other (Figure 9). By some twisting the tail will usually slip out of its skin or sheath. Some tails, as of certain shrews, may need a slight preliminary rolling-pin treatment with the handle of a knife on the table to soften them and loosen the skin a little. Stronger tails may be slipped through the points of a forceps, or gripped between two sticks held in the hand, or squeezed between the cleft of a split stick tied together at one end. Hairless tails

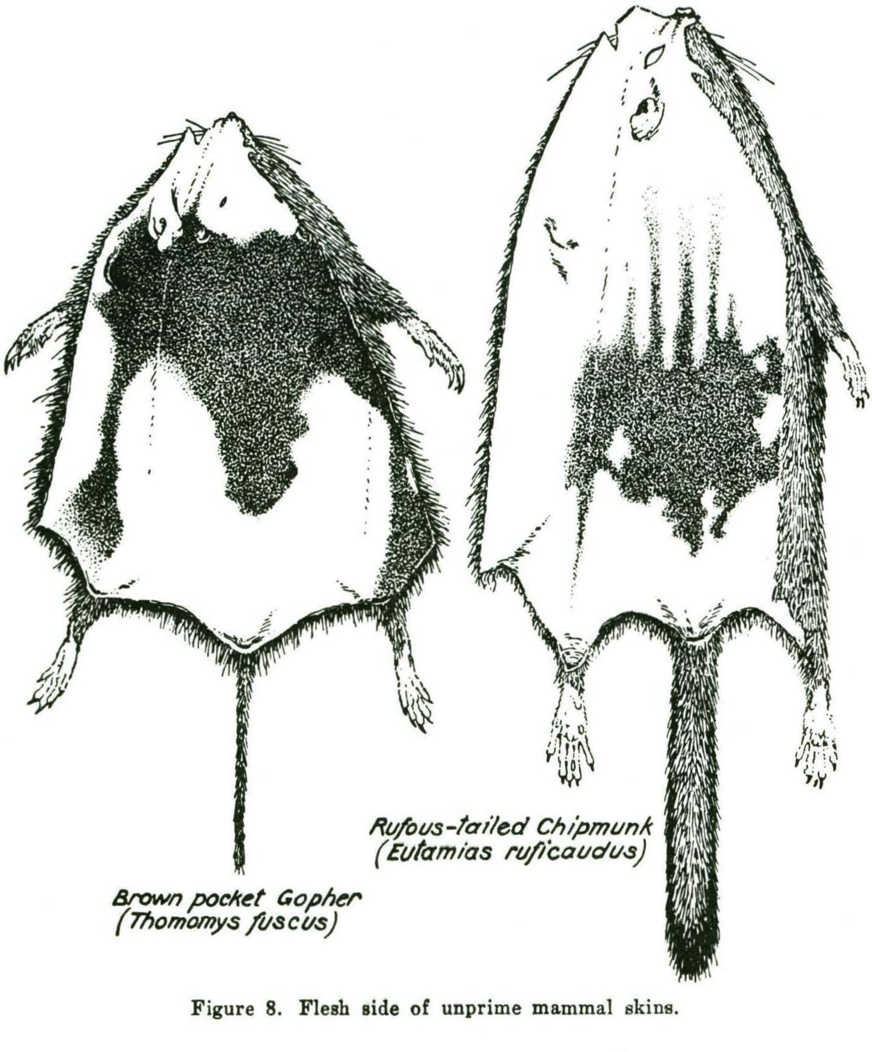

Figure 8. Flesh side of unprime mammal skins.

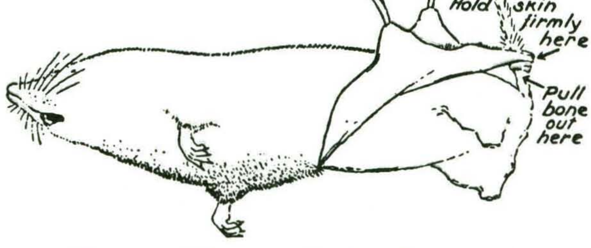

Figure 9. Skinning tail of small mammal.

like that of the muskrat, and heavy muscular or fat tails like those of the skunk, porcupine, or beaver, must be split on underside. In fact, any tail of an animal larger than a squirrel should be split open to remove fat, etc., and enable it to dry more readily. The tail being free, work the skin gradually free from the body. For the convenience of beginners, and, in the case of large skins, of anyone, the body may now be hung up by a slip knot around the hips or legs, so that the operator may have both hands to work with. Dust on plenty of sawdust from the time the opening cut is made, to absorb blood and grease, and to afford a better handhold on the skin. Avoid stretching the skin. Cut off the fore legs and continue peeling the skin down to the head. When the bases of the ears are reached, these may be cut off with the scalpel as close as possible to the skull. The eyes will soon be seen, and the membrane attached to eyelids should be cut through with the scalpel, but with great care not to cut the eyelid itself. This necessitates cutting deeply with the point of the knife. If the animal is large enough, thrust the finger into the eye from the outside and cut against the finger. This will ensure proper care. Cut around the lips close to the skull at the inner edge of the gums; and free the nose by cutting through the cartilage near the tip (Figure 10). The remainder of cartilage at tip of nose may usually be peeled out by the thumb or finger-nail, or by scraping with knife, and it should be carefully removed or the mammal's nose will shrink to a peak when drying.

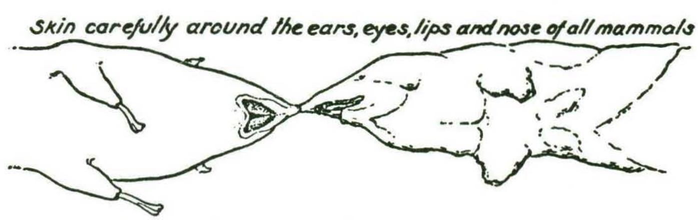

Figure 10. Skinning head of small mammal.

The skull then comes out and should have a label attached and be hung up to dry. Scrape or pull off any bits of fat or meat on the skin, using sawdust as an absorbent. The flesh and fat may be removed from the skin of body and legs with a knife or notched scraper, but around the eyes, ears, cheeks, and nose, the scissors come into play. Hold thumb or forefinger on fur side and stretch skin over it, then shear away flesh by holding the scissors flat on the skin. Avoid cutting any wrinkles or through roots of hair. Bring lips together with a stitch or two. Cut or scrape away any bits of meat from the stumps of the leg bones and skin these as far down as the skin will slip conveniently without tearing. Sponge off any blood or grease from the hair, and rub dry preservative into all parts of the damp flesh side of the skin.

Make a stretcher from a piece of thin board, cardboard, or corrugated pasteboard, or use a stretching frame (Figure 11) about the width of the cased skin as it lies flat, and pull the skin over the stretcher, fur side out, to dry (Figure 12). Animals larger than a squirrel should be put on the stretcher with the flesh side out for a day or two. When the skin is partly dry, but not brittle, turn it right side out and hang it up to complete the drying.

Sumner and Swarth (1924) described a method of stretching small skins uniformly for use in accurate determination of colour tones by instrumental means, the flat skin being laid out on a block and small pulley weights attached to the edges of the skin. However, they seem to have prepared many of their skins by first measuring the animal in the flesh, then stretching the skin slightly by hand while it was being pinned to the drying-board. The skin was pinned at eight points—tip of nose, extremity of each of the four feet, one at middle of each flank, and one at tip of tail. An important part of this technique consisted in thorough removal of grease from the skin by means of benzene.

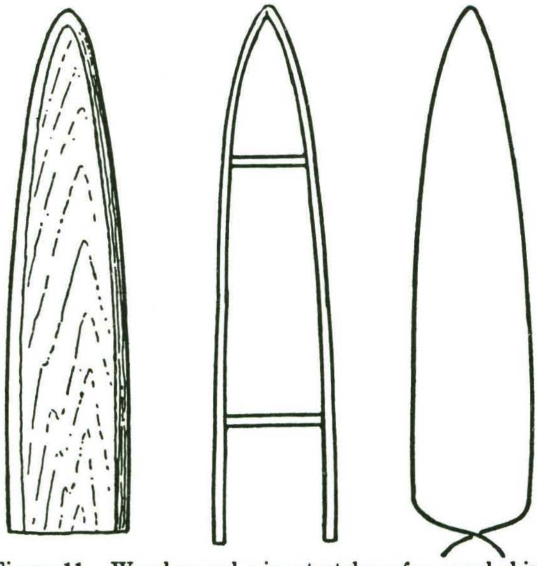

Figure 11. Wooden and wire stretchers for cased skins.

The writer as well as many other collectors have pinned or pegged out skins to dry in emergencies, but have not found the method well adapted for small skins as they break and tear easily when being examined.

Great care should be taken not to overstretch a skin, and to dry it as small as possible without allowing wrinkles to form. A skin which has shrunk in the natural process of drying can be moistened and stretched at any time, but when a skin is once over-stretched, it is almost impossible to reduce it to natural size again. If the skin is allowed to wrinkle, the sides of the wrinkles may come together, preventing rapid drying, and local decomposition may set in and the hair slip off in the cracks. *Where preservatives are not available, the skins of all animals should be dried flesh side out.* Small skins dry very quickly, and had best be left as they dry, until they can receive laboratory attention. If the skin is properly cleaned and stretched it will need no other attention, except to keep it out of the reach of mice, moths, and dermestes.

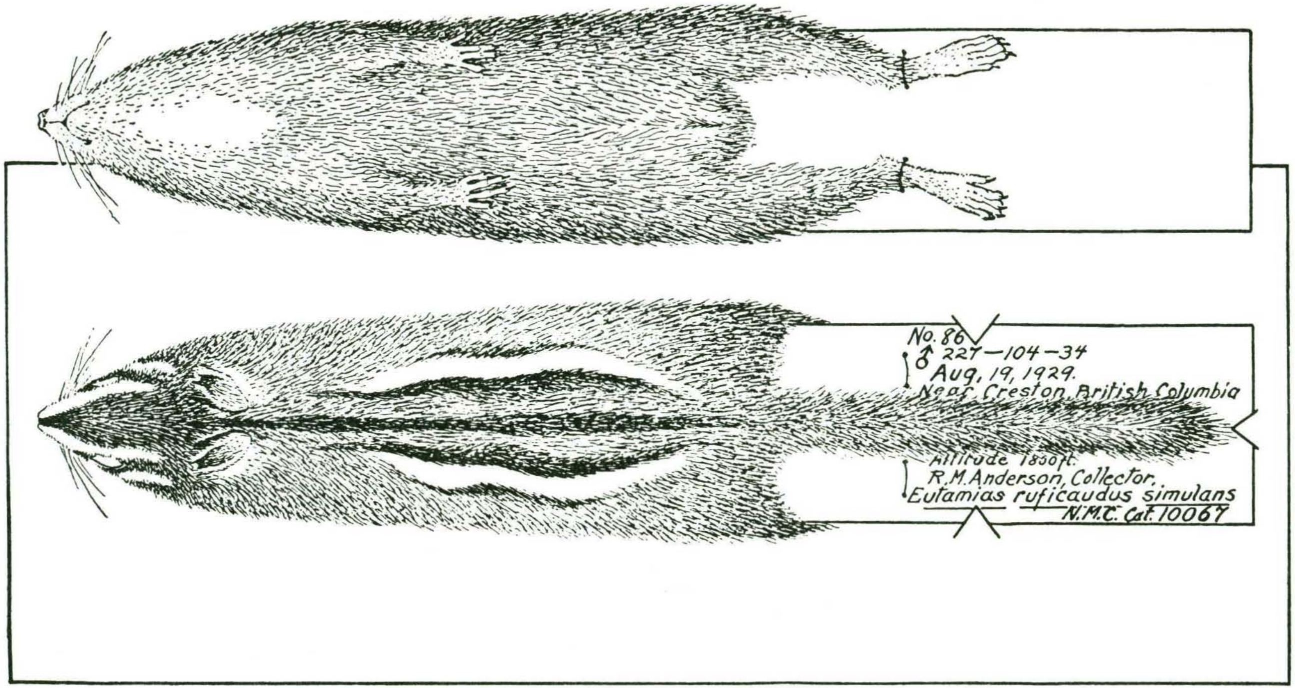

Figure 12. Cased skin of chipmunk (ventral and dorsal views).

Where many skins are handled, professional trappers usually keep drying-boards or frames on hand. Wire stretching-frames are inexpensive, may be easily made or purchased, and may quickly be adjusted to any size of pelt and allow space for a circulation of air between the sides of the pelt, thus hastening the curing process. Another great advantage of the wire stretcher is that the springy sides need only to be compressed and the dried skin slips off without sticking or tearing the hide or the fur. The average collector, who only puts up an occasional cased skin, will usually find it less trouble to improve a stretcher from a piece of packing box, shingle, split cedar shake, or bent willow saplings, than to keep on hand elaborate equipment that is seldom used.

An adaptation of the method of "casing" skins used by commercial trappers as described by the writer in the first edition of this work (1932, pages 45-48), was described as a practical means of getting mammal skins from persons who had neither the time nor the inclination to "make" the conventional type of "scientific or study skin." Such skins are as useful as any other "make" for the purposes of mounting, and to all intents and purposes known to the writer are perfectly suitable for study purposes. If the owner insists on having his skins uniformly made it is quite feasible to relax the cased skin and make it up in the orthodox style, an operation which has been commonly done in the past. If pasteboard is used for a stretcher it may be left inside the skin and the necessary data written on the bottom (Figure 12). If badly soiled, the stretcher may be replaced by a clean one. It has also been suggested that if a permanent mounting card be cut from transparent sheet celluloid to replace the original cardboard stretcher the hind feet and tail may have the under surfaces examined without removing the stitches binding them down.

Mr. Charles Elton, Director, Bureau of Animal Population, University Museum, Oxford, England, has been carrying on extensive experiments in preparation and storage of flat or cased skins, and referring to the method described by the writer in first edition of this bulletin (1932), writes as follows (1938, pages 244-245):

> "The following extensions of this technique have been made. Suppose one has cased a mouse skin: instead of tying a label onto the animal, one leaves sufficient of the stretcher card behind the hind legs to allow the data to be written upon it, and the hind legs and tail to be secured to the card with thread. The base of the card is cut to a standard width that is considerably wider than the mouse, the width used for small mammals, such as mice, is 4 inches. The whole mounted specimen with its card is put inside a cellophane envelope, 4·25 x 11 inches, open at both ends so that the mouse can always be slid in and out with the grain of the fur. The resulting product is a flat specimen of which both sides can be examined without removal from its envelope. It can be removed in an instant if desired, the hind legs and tail are safe from injury, and the records are securely attached to the skin and are visible at a glance. The mounted specimens can then be stored in cabinet drawers or boxes, and classified with guide cards or individually distinguished. This system has especial value where large numbers of skins are being collected for ecological or genetic study, but it may prove also to be of use to the museum expert, provided he does not insist on having round skins for comparative work. The cased skin is inevitably wider than the animal, but this point appears less serious when it is realized that reliable measurements can in any case only be made on the body itself. . . . Casing skins is at least as speedy as any other method, but there is a potential waste of time in cutting cards to the exact size required in each instance. This difficulty is overcome by having a set of flat metal gauges by whose aid the right size of card can quickly be found. Each gauge has a line drawn down the centre marked at

centimeter intervals, that facilitates cutting the base of the cards to the right depth to clear the end of the body. The gauges are held together at their bases by a central rivet, and they pack into a small space . . . The cost of cards and cellophane envelopes is quite small, and is balanced by the great saving in overhead costs of storage and the convenience of reference."

Arthur and Ruth D. Svihla of the Department of Zoology, University of Washington (1939, page 111), have recently tried out the above method in the field with a few modifications and found it time-saving, convenient, and satisfactory for such small mammals as mice, voles, shrews, and weasels. It was found that by making the slit from leg to leg along a line midway between the anus and the urethral opening, more skin support is given the base of the tail and a smoother ventral line is obtained. They use corrugated pasteboard from ordinary packing cartons for the bodies, it being stiff enough for ample support and porous enough for quick drying, and one quickly becomes adept in approximating the correct width and trimming the cardboard to shape. Two leg wires only are necessary, one for each side of the body. These can be bent slightly at the feet so that they turn in, holding the feet to the ventral side of the skin. This ensures their protection and prevents them from being broken off. A single tail and body wire was cut a few millimetres longer than the total length of the animal, inserted through the base of the nose and passed along the mid-line of the body to the tip of the tail. The wire projecting from the nose was then clamped down ventrally holding the cardboard and skin firmly together.

The label can be sewn to the cardboard so that the free end just tucks under the skin, and the data can also be printed by hand very easily on the cardboard body after the specimen is made up.

Whether the collector uses the wide based stretcher described by Elton, or the ordinary shape used by trappers is a matter of individual preference. In either case the data can be written on the base of the card, and a more formal label also attached if desirable. If a wide-based card is found necessary to hold the specimen in place inside of the cellophane envelope, it may be slipped on top of the original stretcher at any time, or the old stretcher slipped out and a new one substituted. The writer recommends degreasing the skin with carbon tetrachloride before attaching a clean museum label. If a clean, degreased space is visible on the flesh side of the skin, large or small, it is well to print the catalogue number with a pen and India ink, to avoid mixing specimens if the label is removed.

Standard Study Skins

In most museums and large private collections the skins of mammals smaller than a raccoon are made up as study skins or so-called "scientific skins." Medium-sized and large skins are usually cut open and spread out. The extra trouble involved in making up "study" that is "stuffed" or "round" skins instead of the simpler "cased" or "flat" skins is counterbalanced by the convenience of comparing uniformly made skins, as most museums use the conventional make of study skins for scientific reserve series. Flat skins are more apt to have the tails and legs torn off in handling.

For purpose of explanation the writer has selected a short-tailed shrew (*Blarina brevicauda*). After taking measurements, writing labels, and recording data in field catalogue, the collector should plug with cotton the mouth, nostrils, and shot holes of the specimen if blood is apt to flow. If the animal has been dead for some time so that the blood is congealed, it is usually not necessary to plug holes. Lay the specimen on its back, part the fur along the mid line of abdomen, and make the opening cut from about the breast bone to the base of tail, making the cut run to one side of genital organs and perineal regions (Figure 13). After starting the

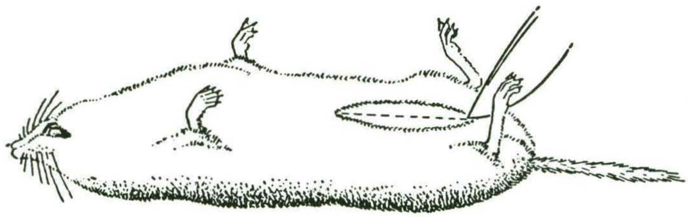

Figure 13. Making opening cut for study skin.

incision, if the skin is loosened with the handle of the scalpel, and the cut is made with edge of blade up, there is less likelihood of cutting off the ends of hairs, and of penetrating the abdominal cavity. If the abdomen is perforated, and juices run out, smother them with sawdust, and keep any exposed flesh coated with sawdust from beginning to end of the operations. The free use of sawdust will keep the fur clean and prevent hair sticking to the body, as well as giving a grip on the slippery surface of the skin.

Use the fingers or flat end of scalpel handle to loosen the skin from the flesh, and work the skin loose from the knee and upper leg. Grasping the foot, thrust the knee-joint upward, disjoint it with knife, scalpel, or scissors (Figure 14), and work the skin as far back as it will go—usually to the heel in small mammals. Some collectors save time here by cutting

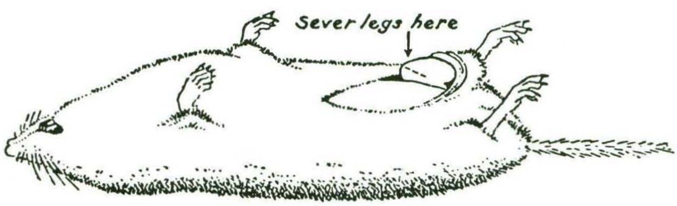

Figure 14. Skinning legs of a small mammal.

the leg off at the heel joint, but pay for it later by not having the bone to wrap the leg filling around. Clean the flesh from the lower part of the leg. The muscles usually peel off easily by slipping the blade along the bone and severing the tendons near the joints. Skin the other leg in the same manner.

Work the skin loose around the base of the tail and slip the tail vertebra out from the skin or sheath as described on page 48 (Figure 9). Sometimes a tail is broken in a trap, or is severed by a shot, and sometimes too strong a pull will bring out part of the tail. Squeezing and manipulating the adhering part will generally enable it to be drawn out by its tendons. Otherwise the skin may be split for a short distance on under side and sewn up afterwards.

With both hind legs and tail free, the skin is rolled back, the forelegs cut off, and bones cleaned of flesh, head skinned, and skull removed as described for "cased skins" (Figure 10).

In removing the skin never pull it until it stretches, but manipulate the skin gently, using finger-nails and the blunt end of the scalpel, cutting or scraping away any strands of fascia that hold the skin to the carcass. The free and frequent use of sawdust or cornmeal on the skin will greatly facilitate this work.

Clean the skin as described before, washing it with soap or soda in water or, if very greasy, with a solution of gasoline, alcohol, and turpentine. Squeeze out the moisture, but do not wring the skin and thereby stretch it out of shape. Dry the fur, rubbing in fine sawdust or cornmeal, repeating the operation as often as necessary, and beating out the dust.

The skin and attached bones being cleaned, the lips may be brought together with a few "surgeon's stitches," tying a knot for each stitch, and cutting off the thread. A continuous stitch with one thread, such as a "ball stitch," will bring together the wrong parts and give the mouth a twisted appearance. The first stitch should be made through the inside of the lower lip, from the posterior edge forward, and coming out inside the edge of the exposed part of the lip. Draw the thread through a point at the middle of upper lip and nasal septum, tie a knot, and cut the thread. For a mouse two more similar stitches, one on each side between the middle and the corners of the mouth, will be sufficient, but with larger mouths such as those of squirrels or rabbits five stitches are better, as it is not desirable to have the cotton filler show between the stitches. With the stitching done from the inside in this way, the thread should not show on the outside of the finished study skin. Many collectors merely bring the lips together with a triangular stitch (Dice, 1932) through each side of the upper lip and once through the lower lip, tying securely. Any large cuts or holes in the skin should be sewed up from the inside using an overhand stitch.

Shrews are difficult to handle, being small, and having a very long snout and upper mandible, with a correspondingly long upper lip, but a short lower lip. The ordinary method of stitching distorts a study skin, and a better method is to run a thread through the two sides of the upper lip about one-third of the distance back from middle angle, bringing them together, thence back through the inner edge of the lower lip, thence through the middle angle of the upper lip, drawing the four perforations gently together and tying the two ends of the thread together (Figure 15).

Poisoning the Skin

Put a little arsenical soap in a saucer and mix it with water, or, preferably, keep soap in a wide-mouthed receptacle so that a depression may be made in the top and lather worked up with a wet brush. Paint the whole fleshy inside of the skin with arsenical soap, working it well down to the feet and tail sheath and around the head. If a brush is not at hand, a wisp of cotton or bit of cloth wrapped around the tip of a small stick may be used. Dip the wet skin into dry sawdust and shake off at once any sawdust that does not adhere. The poisoned skin will be coated with sawdust and if handled gently very little arsenical soap need touch the hands. The water with the soap makes the skin soft and flexible at all points, so that it can be easily shaped. Beginners, and any who may be interrupted or delayed in the skinning and cleaning process, may find that a small skin has dried in spots. If dry preservative is being used (arsenic, alum, borax, saltpetre) the skin should be sponged or moistened with a wet rag or bunch of cotton dipped in water, or the skin may be worked up with wet fingers.

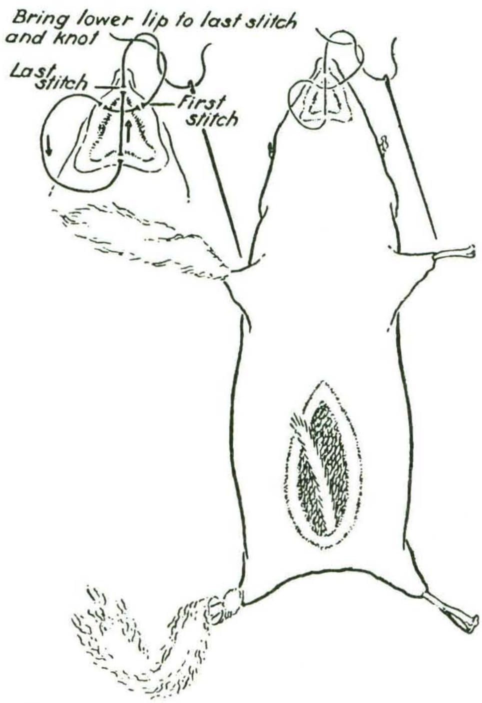

Figure 15. Sewing up mouth of a shrew.

Many collectors faced with several specimens prefer to do all the skinning at one time and stuff the skins later after the whole lot have been skinned and cleaned. This plan has some advantages in the way of efficiency, and saves time. The plan is excellent in hot weather when no ice is at hand and when the difference of an hour or so will start specimens on the way to decomposition. The skins may be rapidly removed and placed in a tin box with tight cover, or a box with damp sand or sawdust in the bottom. When the bodies of the specimens are to be used as food (as with ducks, grouse, plovers, etc.) it is better to finish the skinning operations and get the meat out of the way before the poison is brought out. When operations are conducted in this wholesale manner, great care should be taken that the skins are marked or labelled so that in mammal specimens the skull goes with the right skin, and that the birds are properly sexed. If several bodies are on hand at once, there is a very great danger of mixing them up. A certain amount of bookkeeping is necessary if the specimens are to be of scientific value, and too much should not be left to memory.

Dry powder (arsenic, alum, borax, saltpetre) may be dusted on and will stick to a moist skin, but if the skin has become dry it should be moistened by sponging with a wet rag or bunch of cotton. A small dry spot may be softened by dabbling on a little water with the finger and rubbing it in. The powder may be dusted on with the point of scalpel, with a small brush, a rabbit's foot, or a bit of cotton on a stick. Rub an extra amount into scalp, feet, and base of tail, and shake the surplus back into the poison can. If the animal is fairly large and the skin does not dry quickly, the soles of the feet should be slit, the flesh removed, and a little preservative put in. The slit may be closed by a stitch or two.

After finishing the work, if poison is used, the hands should be thoroughly washed and the nails cleaned with a soft stick. Medical students often rub the finger-nails full of vaseline or cosmoline before beginning a poison-pickled or malodorous dissection, and subsequently wash the hands with hot water and green soap.

Filling the Skin

Before turning the skin right side out, the leg bones should be wrapped with cotton, both to shape the skin and to prevent the bones from adhering when drying. A small, flat sheet of cotton is peeled off the roll, and one end wrapped tightly around the stump of bones, the thicker end forming the upper part of the legs and later lying alongside the body filler (Figure 18*d*). If the upper bones of the leg have been cut away, a leg

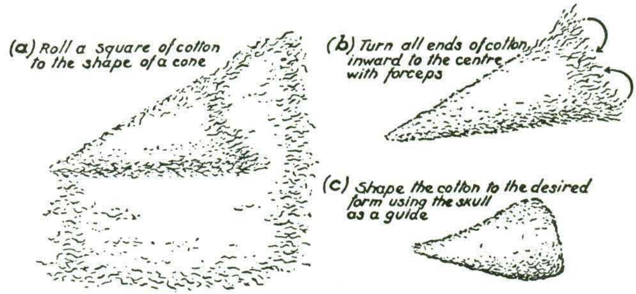

Figure 16. Making artificial head for a small mammal skin.

wire should be wrapped with cotton batting. The wrapping should not reach to the tip, but begin at the position of heel or wrist. The end of the wire is pushed into the foot, preferably on the top of the foot or along its side.

Turn the skin right side out. The first step is to fill the skin of the head. Peel off a small square of fine-fibred cotton and roll it into the shape of a cone (Figure 16). Twist and squeeze it hard by pushing it with fine pointed forceps, a piece of stiff wire, or knitting needle into a loop made by the forefinger and thumb. Turn all ends of cotton inward to the centre with forceps and shape the cotton to the desired form, using the skull as a guide. Skulls that are angular or odd-shaped like a rabbit's should be imitated as closely as possible to make a good looking "skin." Some collectors think that equally good results may be obtained with shrews by making head and body filler in one piece, but the writer's experience is that the pointed head of the shrew cannot be made firm enough in that way. The tail must now be wired. Splinters of tough wood or bamboo are often convenient substitutes for wire. A wire may be straightened by putting one end in a vise or around a nail and pulling strongly, or by stretching the two ends apart while holding each with a pair of forceps. The wire must be small enough to go to the tip of the tail skin, rigid enough not to bend easily, and proportionate to the size and durability of the skin. Cut a wire the length of the tail plus a little more than the length of the opening cut in the body skin. Wire for tails that run to a very fine point should be tapered with a file for an inch or two. The terminal part of the wire should be slightly roughened with a file so that the cotton wrapping will not slip. Wrapping the tail wire for a long haired tail is not very difficult, as the only important thing is to cover the wire with cotton at all points. Wiring a smooth, short-haired tail takes some practice, as the artificial tail must be smooth and tapering; its imperfections will be glaring when the skin dries down upon them. The tail of the jumping mouse may be taken as a good example. Flake off the thinnest possible wisp of fine-fibred cotton, wet the wire so that the cotton will stick, and beginning at the tip wrap the cotton firmly around the wire, twirling the wire with one hand, and gradually thickening the wrapping with the other, following the natural tail as a model (Figure 18*c*). If the wrapping tapers too much at any point it may be built up by wrapping on flat, slender wisps of cotton, but care should be used to wrap tightly or the cotton may slide into a bunch on the wire when insertion is attempted. If this happens, the best thing to do is to wrap a new tail, as forcing in a lumpy tail is apt to pull off the end of the tail skin. If the tail is spindle-shaped, as in the star-nosed mole or some shrews, wrap the wire with cotton to imitate the natural shape of the tail, and cut a slit underneath the basal part of the tail in order to allow the wired tail to enter. The slit may be sewn up with a few stitches. Painting the wire tail with arsenical soap makes it slip in easily, but if dry preservative is used, the tail may be wet and rubbed in the powder before insertion in the sheath. After working the wire into the tail properly and seeing that the skin is not twisted, the body filler may be prepared.

It is important that all the body filler should go in one smooth, firm roll, because if separate bunches are put in the skin, it will be sure to have a lumpy appearance when it dries and shrinks. Irregularity is much more noticeable in a mammal skin than in a bird skin, as the imperfections of the latter are disguised by an overlapping sheath of feathers. Roll an oblong piece of cotton until the circumference is roughly that of the body. The cotton body will be a little larger than the skinned carcass, as the body becomes more or less deflated after death. The skin will also shrink down on the body filler to some extent while drying. The intention is to make the stuffed skin look like the animal in life. Turn both ends of the cotton inward to the centre with forceps, push the ends inward until the exact body length is secured, using the skinned body as a guide (Figure 17). The anterior end of the tail wire may be bent up a little and slipped inside the cut along the abdomen.

Grasping the cotton head-filler firmly at each side with the forceps (Figure 18), insert it through the opening cut and make it go to the extreme tip of nose. Follow this with the cotton body, shoving one end firmly into the hollow at the base of the cotton head and tucking the rear end of body neatly around the hips. Arrange the legs in natural position, the loosely wrapped ends lying flat against the body filler (Figure 18).

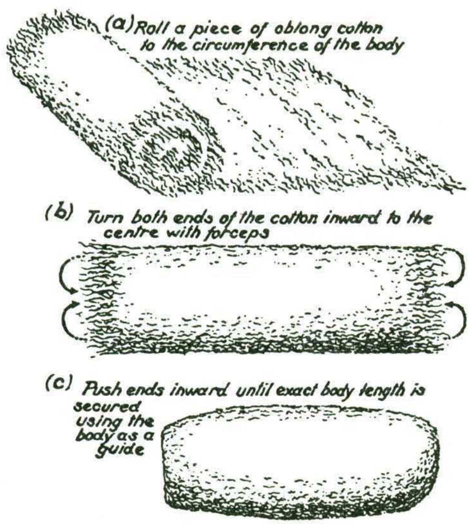

Figure 17. Making artificial body for a small mammal skin.

Before sewing up the skin, see that the ends of leg cotton lie parallel along the body, adjust a thin layer of cotton over the end of the tail wire, and put a little extra padding where necessary around the base of the tail, sides of the shoulders, etc. A common fault is to have the hips break off abruptly and leave a shrunken space around the base of the tail. Tie the end of the thread into the skin at the first stitch, as a knot often pulls through. A common practice is to catch a little lump of loose cotton in a slip knot at the end of the thread to make a stop knot, but care should be taken that this does not form a lump under the skin. Sew up the opening, beginning at either end of the slit, using the baseball stitch (Figure 19) from one side to the other, putting the needle from inside of skin to outside, and taking care not to catch any tufts of hair in the stitches. When all the stitches are in place, gradually draw the edges of the opening together, carefully take up the slack in the thread, and make the end fast by an extra stitch and knot. It is generally more unhandy to draw each stitch tight as it is made, as the overlapping feathers or hairs get in the way of the next stitch.

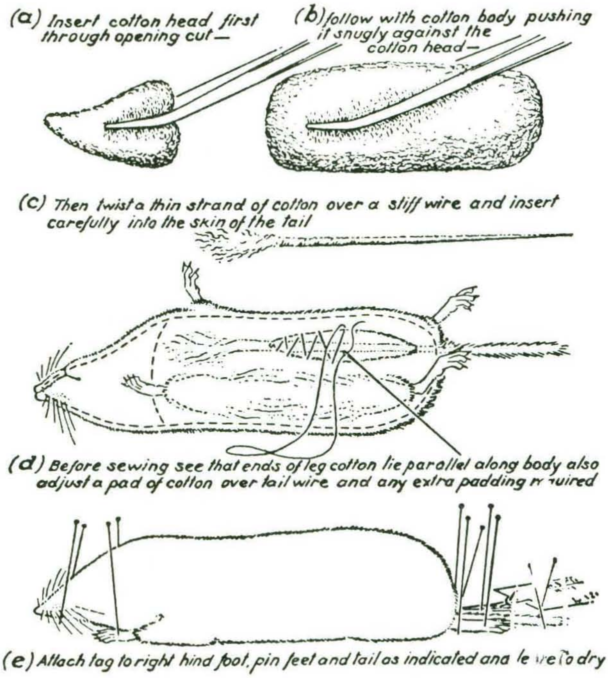

Figure 18. Filling a study skin of a small mammal.

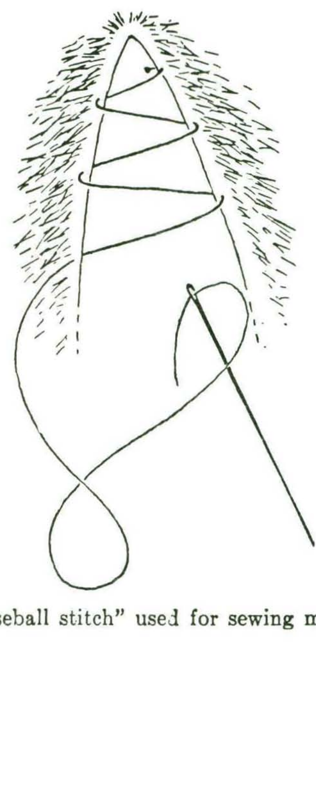

Figure 19. The "baseball stitch" used for sewing mammal and bird skins.

Laying out Specimens for Drying

In order to dry properly, the skin should be pinned out on a board. If obtainable, sheets of pressed cork, such as are used by entomologists for pinning out insects, are superior to anything else for small mammals, but any soft board that pins can penetrate will do.

The stuffed skin will probably be somewhat rough and distorted, and should be laid out as nearly as possible in the shape and size of the dead animal. It is convenient to keep the skinned body at hand until after the laying out of the skin, but the length of body and tail can be taken from the measurements made at the start. Lay the skin beside the skinned body and gently compress or elongate the skin with the fingers so that the lengths of tail and body correspond with those of the carcass. The writer's method is to anchor the skin in place by a pin through the base of the tail, then to place a pair of pins crossing over the middle of the tail to hold it in line with the body. Bring the tip of the nose to its proper place, and draw the front paws into line, parallel with the sides of body, pinning them in place by one pin through each paw. The hind legs should then be drawn back into place, parallel with body, and pinned with back of foot up. One other pair of pins should be put in the board at the outer side of each

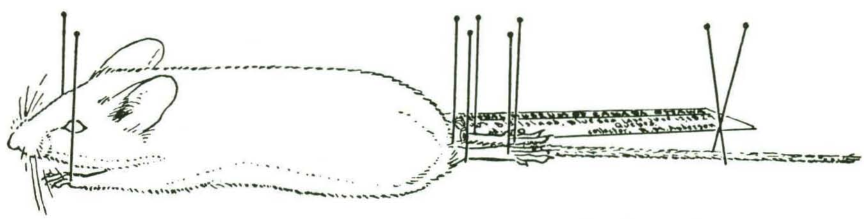

Figure 20. Pinning out a small mammal skin for drying.

foot near the heel to keep it parallel with the body (Figure 20). If necessary, pins may be put between the toes to spread them. This is more desirable with webbed toes or toes with fringed edges, such as those of the water shrew. If the head is properly stuffed it need not be pinned, but if the nose has a tendency to spring up, it may be held in place by a pin set at an angle along each side of face. Be careful that these pins do not leave a groove in the damp skin.

Tie the labels with the complete data to the right hind foot, making a secure double knot just above the heel, so that the thread cannot slip off (Figure 20). Never put on a label with a slip knot or half hitch as the label is apt to get lost. It is best to put a label on the skull at once and hang the skull up, but it may be pinned for drying on the board with the skin. A pin should be run through the space inside of the cheek bone, but holes should never be made in a mammal skull. Some well-made skins of different species are shown in Figure 21.

Burt (Mammals of Michigan, 1946) gives suggestions on collecting specimens of the groups found in Michigan, a list of representative habitats, and (pages 32-44) on preparation of specimens. His method of preparing small mammal skins differs from the preceding methods little more than

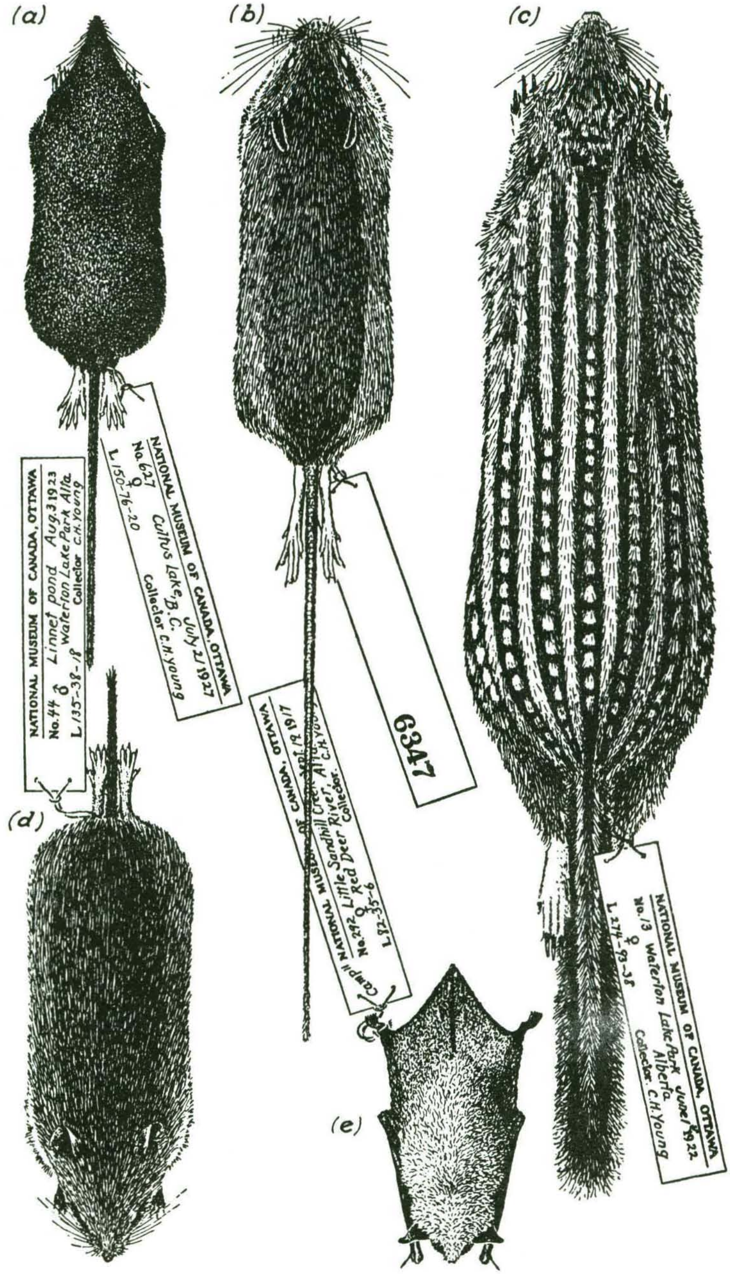

Figure 21. Examples of well-made small mammal skins: (*a*) water shrew, ***Sorex palustris;*** (*b*) jumping mouse, ***Zapus princeps;*** (*c*) striped ground squirrel, *Citellus tridecemlineatus;* (*d*) red-backed mouse, *Clethrionomys gapperi;* (*e*) Say bat, *Myotis subulatus.*

in cutting off the bones of hind leg just above the ankle and the bones of fore leg just above the wrist; cutting four straight leg wires about two-thirds the length of the body, to be wrapped in cotton and the uncovered tips forced into the feet just under the skin of the sole. This may stiffen the skin a bit more than by leaving in the wrapped leg bones, prevent a leg from being pulled off by a dangling label, and make the skin more durable for class use. This method involves the use of four additional wires in the legs. If the wires are not available a narrow strip of bamboo or hard-wood slivers might be substituted, but the collector should be familiar with the method of leaving the leg bones in place.

If the skin has been torn before beginning operations, it may be better to remove the body through the original gap instead of making a new cut. Keith Reynolds, of London, Ontario, recently sent the writer skins of short-tailed shrew (*Blarina brevicauda*) and star-nosed mole (*Condylura cristata*), showing a method used by A. A. Wood, of Strathroy, Ontario, on shrews and moles, species which are prone to decompose rapidly in hot weather. When the hair on the abdomen has started to slip or become loose, he finds it better to make the opening cut from middle of throat along the breast to tip of sternum. With careful handling this will to some extent prevent the loose hair from rubbing off while the animal is being skinned and the opening cut sewn up. The same condition is less frequently found in trapped meadow mice (*Microtus* group) when the stomach contains a large amount of partly digested, soft, green vegetation. (*See also,* use of alum on skin in such cases, p. 11).

Preparation of Skulls in the Field

The skull is removed from the carcass by disjointing carefully at the neck. Cut off only the largest muscles in medium-sized species, and remove nothing but eyes, tongue, and brain from the skulls of small species. Small bony processes are apt to be cut or broken off in the field and a certain amount of dried tissue protects the skull from such breakage. In small skulls poke out part of the brain with a wire or tooth-pick, because if the whole brain dries in place, it may swell later when the skull is being cleaned and separate the sutures, particularly in young skulls. Large skulls should have all the brains removed. A rough wooden spoon may be whittled out of a stick, or a soft green twig may have the end hammered into a rough brush, and with these the brains may be poked out. After part of the brains have been removed, water may be poured in through the foramen magnum (entering point of the spinal cord) and the skull well shaken and rinsed out before hanging up to dry. The skull should be preserved in as perfect a state as possible, and the foramen magnum should never be enlarged by cutting nor should any bony processes be cut or scraped away.

Skulls may be dipped in cornmeal or sawdust to hasten drying and to keep labels from sticking to the flesh. Do not allow skulls to remain wet, as they macerate and decay when damp, and the jaws and teeth may drop away, so that the numbers cannot be associated with all parts of the skulls. Fractured skulls will usually have the parts kept together by the periosteum and ligaments, but if any pieces become detached they should be tied up in a bit of rag and fastened to the larger parts.

Small skulls dry quickly when hung up. When several skulls are being cared for at once they may be strung on a cord or wire passed through the loops by which the labels are attached, preferably through a special loop made by tying together the loose ends of the thread after the label is fastened. By keeping the loops short the dangling skulls will not get tangled, and any particular skull may be removed without loosening its label. The "strings" of skulls will dry rapidly in the sunshine or near a fire. Care must be taken that they are not stolen by cats, dogs, rats, or birds.

If there is no time to dry skulls, they may be preserved in alcohol, but no alum or formalin should be used, as these retard the cleaning processes. Formalin also decalcifies bones, if they are left in the solution for any length of time. When only a few skulls are to be handled, alcoholic preservation has some advantages, particularly in preventing loss or breakage of small loose skulls. (*See* also Borell, 1938, cleaning by dermestid beetles.)

LARGE MAMMALS

Mammals larger than the woodchuck (*Marmota*) or raccoon (*Procyon*) are too bulky to be stuffed in the field. The skin is removed in a somewhat different manner from that described for smaller mammals, dried flat, with or without the use of chemical preservatives, and tanned later.

It is sometimes difficult to get accurate and consistent measurements. Large mammals should if possible be measured on level ground, in a straight line from tip of nose to end of last vertebra of tail. Never measure around the curves of the back. A tape (preferably a steel one) should be used for all measurements. Measurements taken with a knotted string are uncertain on account of elasticity of the string, even if the collector can remember what the knots mean. Notches on a stick are a bit better, but after all, if a man is going to do scientific collecting he would do well to bring along tape and notebook as well as rifle, cartridges, and skinning knife.

The *three essential measurements* used in scientific comparison should always be taken:

(i) Length, from tip of nose to tip of last vertebra of tail, measured in straight line, with body stretched out.

(ii) Tail, length from base bent at right angles to body to tip of last vertebra, not including hairs at end of tail.

(iii) Hind foot, from tip of hock to tip of hoof, or from back of heel to end of longest claw, the foot being pulled out straight in any case.

Additional measurements (Figure 22) may be taken for the benefit of the taxidermist who may mount the specimen.

(iv) *Height at shoulders*, distance in a vertical line from top of withers to sole of foot or bottom of hoof as the animal would stand in a natural position.

(v) *Chest: circumference* just back of the elbow, measured with a string or tape; *depth* at same point, measured in a straight line between two sticks stuck in ground at withers and brisket; *thickness* in widest part, the distance between spines of shoulder blades.

(vi) *Neck: circumference, depth,* and *thickness* at three places: (A) just back of head; (B) middle of throat; and (C) just in front of shoulders.

(vii) *Abdomen: circumference, depth,* and *thickness* at largest part.

(viii) *Fore leg: circumference, depth,* and *thickness* at three points: (A) elbow joint, near the body; (B) at middle of forearm; (C) at knee joint.

(ix) *Hind leg: circumference, depth,* and *thickness* at three points: (A) at stifle joint (across kneecap or patella); (B) at middle of leg; (C) at hock or gambrel joint.

(x) *Distance from head of humerus to anterior angle of hip* or pelvic bone (form 5 to 7 on chart).

(xi) *Distance from head of humerus to angle of throat* (from 5 to 6 on chart).

(xii) Distance between anterior and posterior angles of hip bones (from 7 to 8 on chart).

(xiii) Thickness of body across pelvis (at points 7 and 8).

The collector will save time in the field, where time is usually pressing, by having in his notebook a copy of a diagrammatic drawing of a large mammal, showing the desired measurements, and by setting down the

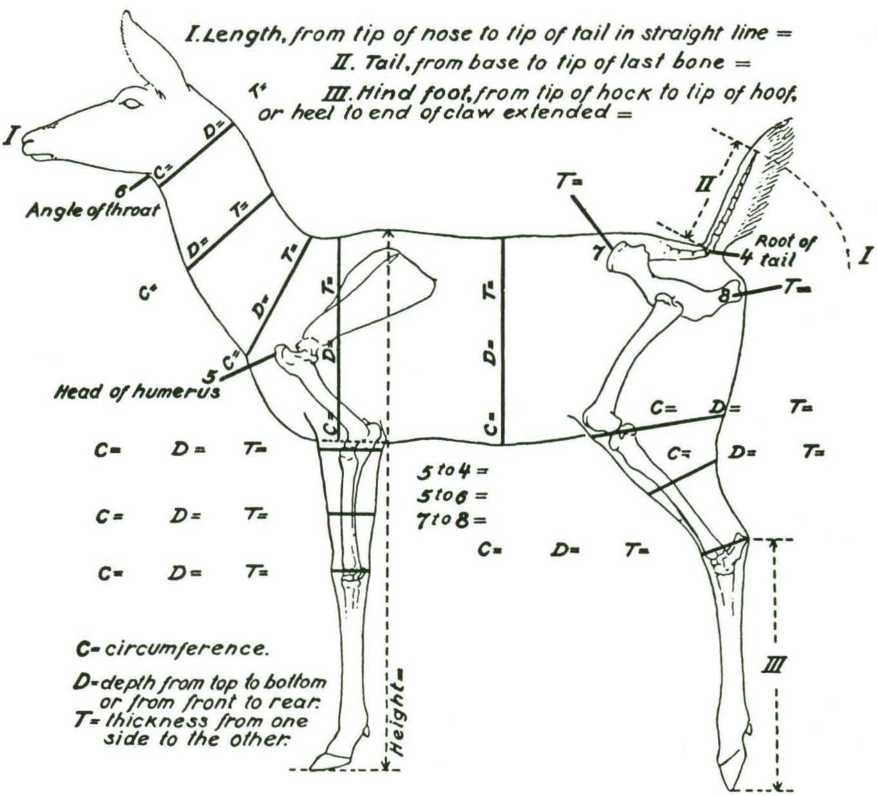

Figure 22. Measurements of a large mammal for mounting.

figures on the diagram as they are taken from the body with the tapeline. Even if the collector does not have such a list of measurements, and has to depend upon memory or reason, he is strongly urged to make a rough outline sketch of the animal he is measuring, and to set down the figures thereon, so that the notes will be sure to make clear to himself and the taxidermist, later on, what the field measurements really meant.

Opening Cuts

An ordinary medium-sized or large mammal is skinned as follows. The opening cut is begun in the median line about the middle of the brisket (breast bone) and the skin worked loose for a little distance on all sides. Then with the edge of the knife up, to avoid cutting the hair or opening the body cavity, run the cut forward to the middle of the throat and backward to the tip of the tail or under side (Figure 23). Make the cut run to one side of the genital organs and perineal region, and avoid cutting through the abdominal wall or the intestines will emerge and possible offensive gas as well. If the hair on the throat is fairly long, the cut may be carried forward nearly to the tip of the jaw if desired, but never through the lower lip. If the lip must be slit, do it at the rear angle of the mouth.

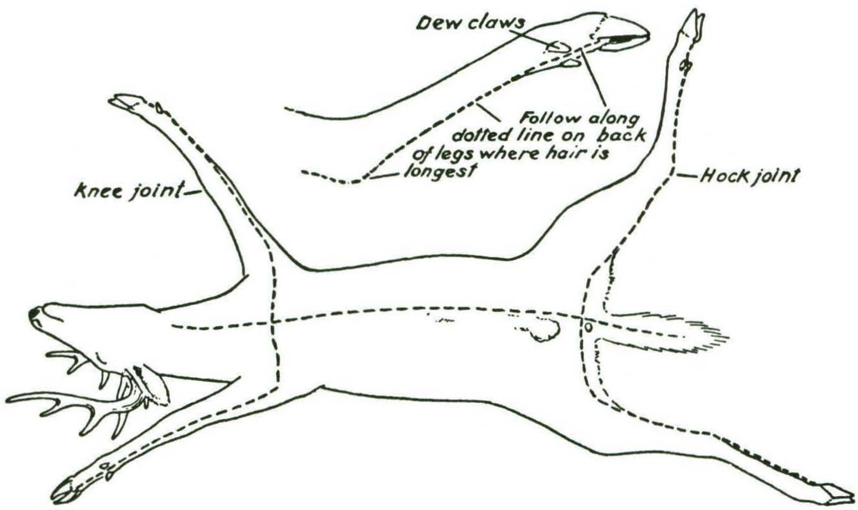

Figure 23. Opening cuts for skinning large mammals.

Then, starting at the heel, run a cut up the inside of each leg to join the median cut; or in hoofed mammals, start at back of hoof and run a cut along the posterior surface of each leg to the hock joint or knee joint as the case may be, thence swing the cut gradually to the inner side of the leg and upward to the median cut. Some mammals, such as bears, must have the skin cut away from the flesh inch by inch, but in others, such as deer, the skin is loose and may be quickly and easily stripped off. A little knife work around the middle of each leg will loosen the skin enough that with a little force it may be pulled loose both ways. The tail and perineal region are dissected loose, severing the rectum well inside the aperture. With some assistance from the knife edge and ripping slashes with the butt end of the skinning knife to tear the fasciæ loose, the rump skin may be grasped firmly and the skin peeled off the back and shoulders with a long, steady pull as the operator walks forward with the end of the skin. Any interfering strings of fasciæ may be severed with the knife. If the animal bleeds much, roll the edges of the skin with hair underneath so that the hair will not become bloody. Trickling blood on the inside of the skin may be mopped up with a handful of grass or mcss. Skinning the head and feet, and other fine points of the job may be done later in camp.

A medium-sized mammal, after the hind legs are skinned, may be hung up by a loop of rope around the pelvis, thus allowing the operator to have both hands free to skin the rest of the body. The big game hunter should carry a few yards of small rope or cod line for such purposes.

If it is desired to hang up a deer carcass out of the reach of predators, split the hind legs at the hock joints and thrust a strong stick through under the tendons of Achilles. Then lift the deer up so that the stick rests on a heavy pole leaning against a heavy tree branch. By giving short lifts and shoving the cross stick up the pole, the carcass may easily be raised until the nose clears the ground. The carcass may also be elevated by a pole tripod, the legs of which are first spread out and then moved in one at a time.

If necessary, a small deer may be packed entire. Make the disembowelling cut as small as possible so that the blood does not run out after draining. Then cut a slit through the skin under the tendon of Achilles (just above each hock joint on hind leg). The front leg is cut through and disjointed at the elbow so that the upper projecting end of the ulna will act as a catch-pin. Then slip each front foot through the cut in the hind leg of the same side so that the elbow joint will catch in the hock incision, locking them together. The packer then puts his arms through the spaces between legs on each side, and hoists the deer on his back with deer's head up and tail hanging down, the abdomen of the deer resting against the man's back. In this way a deer may be carried like an ordinary knapsack load, without the use of any ropes.

The leg bones of large mammals, and the pelvic bones, if possible, should be saved if the animal is to be mounted.

Skinning Horned Heads

Hornless heads of any kind can usually be skinned down to the nose, and the skull removed in the same manner as in smaller mammals. Obviously, the neck skin of an animal can not be stretched enough to go over a pair of horns or antlers. The neck should be skinned up as far as possible through the body cut, and either disjointed just back of the head, or left intact until the head is skinned. In very cold weather or when pressed for time, it is best to disjoint the neck and carry the hide back to camp with the head unskinned. Unless the neck skin is very thick and heavy, as in the buffalo, so that it cannot be rolled up, the writer generally finds the head easier to manipulate after the neck is severed. Frozen specimens should be thawed out slowly, and not be allowed to come into contact with extreme heat, sun or open fire.

To skin a horned or antlered head, a Y-shaped cut is made along the back of the head and neck, reaching a point a little in rear of the horns, with a diagonal cut to the base of each horn (Figure 24). The incision is carried close to the base of each horn, and chiselled deep through the thick skin to the bone of the skull. The skin is cut and wedged away from each horn, worked free from the forehead, back of the head, and the sides of the cheeks, and the bases of the ears are cut off close to the skull. The skull is then disjointed from the neck if this was not done before.

The head is then completely skinned out. Be very careful not to cut the eyelids, as a cut eyelid can never be mended perfectly. This caution is easily carried out by running the finger into the eye from the outside, as a guide to cut against. In a deer, note the deep pit covered with thin skin in the lachrymal bones in front of the eye, and avoid cutting through the skin while working it loose from the pit. Cut both the lips

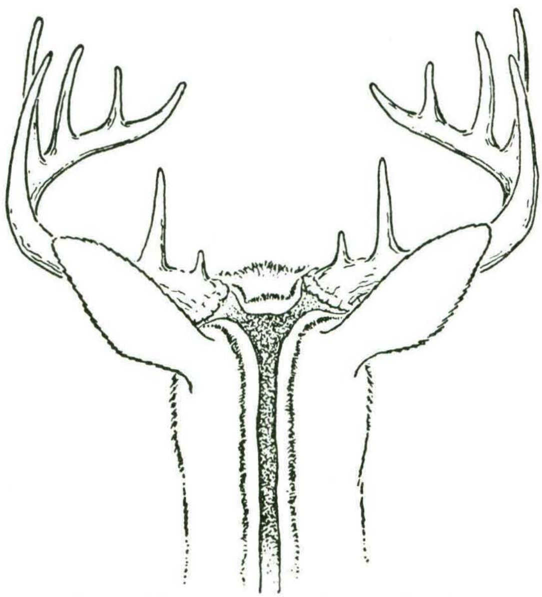

Figure 24. Skinning a horned head.

and the muzzle well back in the mouth so as to leave a wide border on the inner surface of the lip. The skull is then removed through the slit in the back of the neck. In digging out the eyeballs be careful not to punch holes in the thin, bony walls of the orbital cavity.

Heads for Mounting

A scientific collector will always save the skull if possible. If the head is merely to be mounted as a trophy, only the top of the skull connecting the bases of the antlers is needed, as most taxidermists now use papier mâché forms for mounting, both to save time and reduce weight of the specimen. Measurement should be taken from corner of eye to tip of nose in such cases.

It should be remembered that to get a natural pose, a horned head has to be hung on the wall at an angle, with foreneck exposed, and the neck skin must be left long. For an artistic full shoulder mount, the base cut should be made from the withers down along the sides of shoulders and breast point, including plenty of brisket skin. In no case make any slit on the under side of the throat. The back of the neck is slit up from the withers and skinned through the Y-shaped cut. The head-skin, "scalp", or "cape", is fleshed, and well salted before packing.

Special Work on the Head

The nostrils are treated by splitting the median cartilage down to the tip of the muzzle, and splitting the small folds at the tip of the muzzle from the inside. The ears should always be skinned, as the ear cartilage must ultimately be removed in mounting, or the ear will shrivel up like a dry autumn leaf. In skinning the ear, cut carefully about the base, far enough down that the cut will never be seen from the outside of the ear, using the fingers of one hand as a guide from the outside. After getting the cartilage started, push the ear in from the outside as the skinning proceeds and separate the skin from the cartilage as much as possible with the nails and handle of the scalpel.

Skin the ear down to the tip on the back (outside or hairy side). This is usually sufficient unless the skin of the ear shows fat or is densely haired on the inside, in which case the cartilage may be loosened at one edge or the tip and skinned down the inside, freeing the skin completely. In some cases the cartilage adheres so firmly to the skin that the only way to remove it after skinning the hairy side is to make cuts carefully through the cartilage until the edge may be grasped with broad-nosed pliers or between the thumb and edge of a jack-knife and peeled off gently in small segments, aided by pressure of the other thumb on the reverse side of the skin.

"Pocketing" the Lips. The lips should be "pocketed" to allow entrance of preservative, and to form pouches to retain the plastic modelling compound in mounting. If the lips are cut off close to the edge of the hair it will be impossible to make a natural job of the mouth in mounting the animal, and the mouth will have to be closed with unsightly stitches. "Pocketing" is done by splitting the lips all around longitudinally from within by a cut extending to the bottom of the fold formed by the outer skin and the inner mucous membrane, using care not to mar the mouth by cutting through the fold in any place. If the lips are very thick some of the muscular coat may be pared off, but generally it may be dried or cured by preservatives. The finishing touches in cleaning lips are done more easily after the skin has been dried, as the fleshy substances soften more quickly than the skin.

The muscular ring around the eyes should be pared down and in large mammals the eyelids should be split from the inside in order to preserve the eyelashes. Many mammals have bristly hairs or whiskers, the roots of which should not be cut off or the hair will fall out.

Antlers in the "Velvet". With the exception of two small Asiatic forms, the males of the deer family (Cervidae), as well as the females of reindeer and caribou, have antlers that are shed annually. The new antlers begin to grow in early spring, appearing as small knobs which are richly supplied with blood vessels and nerves. These knobs grow very rapidly with deposition of bony matter. The outer skin is very sensitive and is covered with very fine, short hairs known as "velvet". When the growth is nearly complete, a bony ring or burr is deposited around the base of the antler, gradually shutting off the blood vessels, so that the velvet skin dries up and is rubbed off in autumn.

Deer antlers that have nearly reached full size will usually dry without decomposition or shrinkage if hung up. If blood and serum collect and bulge at the tips of the prongs, slit these at the ends, drain out the liquid, and rub in a little alum or salt. If there is any further sign of spoiling, slit the velvet on the under side, loosen it to let in air, and apply preservatives.

Deer specimens in early summer are difficult to handle, as the old hair is very loose and falls off in bunches, and the new antlers are very soft and pulpy and quickly become offensive. However, such specimens must sometimes be collected to make an ecologically correct summer habitat group. When the antlers are very young and knobby, in the soft velvet stage, they may be painted with arsenic water and formalin to keep off flies, and they will dry in time, but will be shrivelled.

The core of the young, growing antler is a spongy, vascular tissue, and if the specimen is to be mounted, the antler may be cut or sawn off close to the head, but first the distance between tips should be measured, so they may be replaced at the proper angle. Split the velvet on the under side and skin it off; then salt down both velvet skin and antler core to serve as a model later. Rowley (1925) recommends puncturing the tips of the antlers with a long, flat awl, driving in wires, and washing and irrigating the inside of the antler several times a day for a week with the following solution:

Water, 1 quart	Alum, 1 pound
Glycerine, 2 quarts	White arsenic, 1 pound
Salt, 1 pound	Formalin, 1 pint

After pickling, rinse off with warm, soapy water, and paint with hide poison (*See* page 74) or strong arsenic water. If a number of pairs of fresh velvet antlers are to be preserved, they may be sawn off and immersed in the pickle. The glycerine in the solution prevents the antler from shrivelling and gives it a natural appearance.

Packing Large Horned Heads. Packing a skull with large spreading horns or antlers is often a serious problem. The skull, prongs, and points may be wrapped with masses of excelsior, grass, or straw, bound on firmly, and wrapped outside with burlap stitched in place. A crate built to the proper size is lighter than a box, but elk (wapiti) heads will need to be boxed tightly in transit as long as elk tusks have a market value for watch fobs, etc. Some collectors save space by dividing a skull vertically down the median line with a fine saw, leaving one antler on each section. This method economizes room to some extent, but is usually disastrous to maxillary bones, nasals, and bones of the lower part of the skull. If the head must be reduced in size, it is much better to saw off each antler, after measuring and recording distances between points. Label each antler and the skull and pack carefully. If necessary the antlers may be reset later at the proper angle, by boring holes for iron rods into the skull and base of each antler. The lower jaw should be wrapped very carefully to avoid breaking the incisor teeth.

Hoofed Mammals

The legs of hoofed mammals are usually disjointed at the hock joints and wrist joints during the skinning operations. If the skin is very large and heavy the legs may be disjointed at the fetlock (lower end of cannon-bone) to make the skin lighter for carrying to camp. After the skin is in camp, the feet must be skinned down to the hoof and the bones removed. The leg incision is continued down to the posterior edge of the hoof and the skin cut, wedged, and pried away from the bone.

With the Artiodactyla (cloven-hoofed mammals) it is usually necessary to run a cut down the back of each of the two large toes. The phalangeal bones (toe bones) should be dug out of the hoof, which is left attached to the skin. Disjointing of the foot bones is usually a somewhat difficult job as the ligaments have to be cut through or gouged loose. A heavy, thick-bladed jack-knife, a small chisel, or a sharp-edged screw-driver will be found useful for this, and after the terminal bone is partly loosened in the socket by encircling it and cutting most of the ligaments from the inside it can usually be worked loose by strong twisting and wrenching motions. The operation of working the bone loose is often facilitated by fastening a stout cord to the bone and to some fixed object to pull against. If the bone adheres too firmly to be removed in this way, a hole may be cut in the bottom of the hoof and the bone further loosened from below and forced out. Care should be taken that sharp-edged instruments do not slip and cut through the skin at the edge of the hoof. If the skin is salted, plenty of salt should be put into the hoof, but if the skin is being dried without preservatives, the skin of the foot should be spread apart with sharpened sticks to allow the air to enter and dry it. Skins should not be dried near a fire, nor exposed to hot sunshine, as a temperature that is hardly too warm for the hand will harden or cook wet rawhide and render it hard and brittle.

Large Mammals with Short Hair

The collector in Canada will find few short-haired large mammals, except horses, antelope, and deer in summer coat. If there is any probability of such mammals being mounted, they should not be opened up the legs nor under the throat, unless absolutely necessary, as it is practically impossible to conceal the seams. With such short-haired mammals, the opening cut is made from the brisket to the tip of under side of tail, and if the animal has horns the head is skinned by the usual Y-shaped cut at the back of head and neck. The legs are skinned by making a cut in the back of each foot from the hoof to dew claws, or accessory hoofs, and the foot is disjointed between the upper end of the toe bones and the leg bones. The legs are disarticulated at hips and shoulders and the skin of the legs is turned inside out, as in skinning small mammals. The skin will strip down easily until the knees and hocks are passed, but will stick tightly to shin or shank-bone. A " leg-iron," resembling a long handled screw-driver, is needed. This has a V filed in the end, with the inside of the V sharp and the outer edges slightly rounded, so as not to tear the skin. By pushing up through the openings in the feet, between the skin and the bone, the connecting fibres are cut, the skin separated from the bone all around, and the bone slipped out. The feet are skinned as usual. Dry salt is rammed down inside the leg skin and the damp salt is shaken out after the skin is cured. This method is used very extensively in skinning short-haired tropical mammals, such as African antelope and gazelles.

Curing Flat Skins

The method of curing flat skins will depend somewhat upon the circumstances in which the collector is placed. Large, flat skins, like cased skins, may be cured by simply drying without any preservative, as is the custom of professional trappers. The cased skin requires less attention, as it is stretched uniformly with no wrinkles to hold moisture. The ordinary trapper or hunter also saves himself trouble in many cases by trimming off the feet, as well as any odd corners, bare spots, and any fleshy protuberances that do not dry easily. For scientific purposes, or for mounting, the skin must be kept whole and unmutilated. A large skin dries more slowly than a small one and needs more watching while drying.

In the woods, the ideal method is to hang the skin on a pole or a rope stretched in the shade, to get free circulation of air and to keep the skin out of the reach of gnawing animals. On the northern plains, or in desert country, it may be necessary to spread the skin on the ground for drying. If the skin is to be used merely for a robe, it may be pegged out, but a better method than making holes is to weigh down the corners of the skin with small rocks, moving them occasionally to allow drying underneath.

Large leg skins in many cases shrink when drying, forming deep wrinkles from which the air is excluded, and in warm weather the skin will soon rot in the wrinkles and let the hair slip. This may be remedied by a few safety pins, blanket pins, or a few stitches to hold the leg skin wrong side out until dried. Split hoofs may be spread apart and ears and lips propped up with short sticks until the skin dries. A little pure alum, or alum and saltpetre, often is useful to rub into refractory soft spots, such as pocketed lips, ears, edges of hoofs, sheath, anal region, and inside of any wrinkles where the skin is not drying properly. Salt is not suitable for such partial or local curing. If used at all it should be used on the whole skin, as salt absorbs moisture from the air and the damp, salty area spreads, preventing the rest of the skin from drying.

In the far north, large skins may be frozen flat after being cleaned, and hung up outdoors over poles or ropes. The dry cold air (from 30 to 50 degrees below zero) will absorb the moisture from the skin in a month or two, and the skin becomes bleached, soft, flexible, and practically dry.

Common salt is useful for temporary preservation of skins, is obtainable anywhere, and is the best medium for poisoning the fresh hides and scalps of large mammals. Salt, the finer the better, may be used for preserving any kind of skin. Table salt is best for small mammals and ordinary barrel salt will do for the larger specimens. Skins designed for mounting are best preserved with salt. It should be applied plentifully and as soon as possible after skinning. One great drawback to salt is that it absorbs moisture very readily, and adds tremendously to the weight of the specimen, thus increasing difficulties of transportation. A raw hide weighing 150 pounds may absorb 50 additional pounds of water and unless barrelled up cause annoyance by dripping brine. Salted skins should be shipped in boxes or bags by themselves, and must be either tanned or remade, using other preservatives, after reaching the laboratory or taxidermist's shop.

Brine, either alone or in combination with alum or acid, changes the texture and certain colours in the hair, transforming bright yellows to a dingy yellow, and dark yellows to purple or brown. Rowley (1925, 115) warns strongly against wetting with brine in any form, the hair of fur-bearers or skins having yellowish hair. If such skins have been dry-salted, and it is necessary to tan the skin for scientific purposes later, several applications of the salt-acid tan liquor must be painted on the flesh side of the skin, without wetting the fur.

Brine has some uses in the field, particularly where other preservatives are not available in quantity. It may be used for pickling some kinds of large skins, such as those of sea-lions, walrus, and large seals, which must be shipped at once without drying, skins of hairless mammals, or skins that cannot be made safe from bugs by other means. It may be used in emergency for shipping bodies, uncleaned bones, and rough skeletons which might otherwise become offensive, as in the case of small whales or porpoises, embryos or fœtuses, large stomach contents, etc., which may be hastily barrelled or soldered up in tins. Brine is a more powerful preservative if boiled to make a saturated solution, but, of course, it should be cooled before specimens are placed in it.

If the skin is bloody, it should be thoroughly sponged with water and the hair combed with a steel comb to remove clots of blood. If very bloody, the skin may be immersed in water, soaked, wrung out gently so as not to stretch the wet skin, and spread out on the ground, flesh side up, to drain. Salt is then spread evenly over the whole flesh surface of the skin and well rubbed in with the hands. The ears, lips, and feet should be well salted, and the skin of head, neck, and legs folded over the body skin, flesh to flesh, and the skin rolled up. The skin should be unrolled the next day and examined for soft spots, where on account of thickness or patches of flesh or fat, the salt has not struck through. Such spots should be pared down and salt rubbed in again. The salt draws water out of the skin and the brine should be poured or wiped off. If the skin can be shipped soon to the museum or taxidermist, it may receive a second coat of fresh salt, and be rolled up in a compact bundle, firmly tied, with hair outside, for shipment.

Hide Poison

If hides must be kept for some time, the loose salt may be shaken out, and the hides dried by hanging them up so as to allow the air to circulate freely through them. Keep them out of reach of rats and mice, as a skin may be ruined in one night. If a skin becomes infested by bugs, it should be first sprayed or painted well with gasoline to kill any insect life in the hair. It may then be dipped in Cooper's Dip, or B.A. Hide Poison, known also as "Venano," which are used all over the world by hide dealers where dry cow and horse hides are kept in quantities. Where these solutions are not available, white arsenic stirred in water and kept stirred while a skin is being dipped, will do very well. Sodium arsenite diluted with water is better, as it penetrates the skin and is soluble. The sodium arsenite solution is made as follows:

Arsenic trioxide (white arsenic, As_2O_3)	¼ pound
Carbonate of soda (washing soda, Na_2CO_3)	½ "
Water	1 quart

Boil for an hour, stirring occasionally (Rowley, 1925).

If the skin softens after dipping, it should be re-salted and thoroughly dried out again.

This formula may be used to poison the exterior of mounted specimens that are infected with moths or dermestes, after spraying or painting the hair with gasoline (applied outdoors). Poison preparations on the inside of the skin help to keep out insect pests and protect the skin itself, but unfortunately there is no substance yet known that will work through the skin and positively protect the hair or feathers. The old method of spraying or brushing the outside of a specimen with corrosive sublimate or bichloride of mercury ($HgCl_2$) dissolved in alcohol is effective enough as far as insects are concerned, but is extremely dangerous and is not recommended, as persons handling the specimen later are very apt to suffer from mercuric poisoning. In dusting such specimens the invisible bichloride powder may be inhaled or absorbed by the skin.

The development of the use of DDT (dichloro-diphenyl-trichloroethane) solution about 1944 for control of various insect pests, including moths and beetles which damage or ruin skins, furs, and woollen fabrics, has been very successful and may supersede some of the older insecticides and repellents. It may be dusted on as a powder, or sprayed in a solvent or emulsion. C. R. Twinn (*in litt.*, 1947) cites experiments by Jensen and Holdaway (1946) who conducted experiments to determine the comparative efficiency of various materials in protecting rabbit hides from larvæ and adults of two species of *Dermestes*. They found DDT to be outstandingly superior to all others. When applied in spray and dust form a single treatment gave excellent protection for at least 371 days, and hides treated while fresh were as well protected for extended periods as were hides treated after they had been dried. Two forms of DDT were used, namely, dusts and sprays. He recommends spraying with 5 per cent DDT in deodorized kerosene, provided that a light application of kerosene would not harm the specimens. DDT itself has been used successfully as a moth-proofing agent on various kinds of textiles. For this purpose it is efficient against clothes moths as well as Dermestid beetles.

Conditions in Canada are such that if fresh specimens are carefully watched in the field and are clean when shipped, hair poisoning is seldom necessary, as the specimens can be treated in the museum before serious damage is incurred. If a large skin, or mounted mammal or head, has been treated with hair poison, it should be marked "Poisoned with ....," for the benefit of persons who have to handle it afterwards.

SPEED AND EFFICIENCY IN TECHNIQUE

Getting into the haunts of big game at the present time in North America usually involves long travel and heavy expense, and frequently when the game is stalked and killed the end of the day is near and time is pressing. For this and other reasons, skins usually have to be taken off in a hurry. The collector of large mammals should learn to do his work efficiently and expeditiously, from the time of planting his bullet in the right spot to make a clean kill to the final packing for shipment.

In regard to skinning mammals, Hornaday (1892, 26) gives advice which the young collector would do well to heed:

"In using the knife do not go at it in a daintily finical way, as if you were picking birdshot out of the leg of a dear friend; for, if you do, it will take you forever to skin your first specimen, and there will be no time left for another. Learn to work briskly but carefully, and by and by you will be able to take off a skin with a degree of neatness and rapidity that will astonish the natives. It is not a dissecting touch that is called for in taking off a skin, but a firm, sweeping, *shaving* stroke instead, applied to the inside of the skin, and not to the carcass. This applies to all skinning operations on all vertebrates, except birds."

The above "speeding-up" is not urged primarily in the interest of increased production, although frequently specimens come in bunches, and if not handled rapidly some may be lost. Under any conditions, a large mammal skins easier before the limbs are stiffened in rigor mortis. In moderate temperatures, such as one encounters on an autumn hunting trip in the north woods, no dire results will follow leisurely methods. Rowley (1925, 112) stresses the difficulty of saving the skin of a large, fat animal under a tropical sun, and states that the hot midday sun shining for an hour on the freshly killed carcass of a lion is sufficient to ruin the skin, particularly the tail, which is invariably fat.

The writer has observed the skin of a bearded seal to blister in a few hours lying in bright sunshine on the ice of the Arctic Ocean with air temperature not over 60 degrees F. The abdomen of a large mammal will begin to bloat with gas soon after death, and though this may not necessarily injure the skin, the flesh will soon become tainted with gas. Even under fairly normal conditions, the collector has to hurry to skin his trophy, cache skin and meat out of reach of predatory animals, or pack them to camp before dark.

Skinning moose, caribou, mountain sheep, and goats in the wilds is heavy butcher's work, and the Canadian collector may have to do the job in snow at timber-line, or in a blizzard at 40 degrees below zero. Extremely cold weather presents problems as well. The dense coat of hair and fur that keeps the mammal warm in life also makes it difficult to thaw out a carcass that has frozen solid. Thawing may take several days in a tent or cabin and the meat may be needed in the meantime. If the shortness of the day prevents complete skinning of a caribou, the animal should be disembowelled, and the legs skinned up beyond the knees and doubled up under the body. If loose snow is kicked up to nearly cover the body, the thick, furry skin will retain the bodily heat until the next day. The writer has cached caribou in this way and found them still warm enough to skin after twenty-four hours of weather 45 degrees below zero. If the entrails are left in over night, the gases of decomposition will taint the flesh of the whole body and sometimes loosen the hair, even at very low temperatures. However, if the collector will learn to do his skinning scientifically he can save time and be fairly comfortable in very cold weather, keeping up his circulation by strenuous effort, warming his chilled fingers and thawing the blood from his frozen knife blade between the skin and the warm flesh as he works. The writer spent 2 or 3 hours skinning his first sheep on a mountain side at 25 degrees below zero, frosting several knuckle joints in the operation, but after some experience shot, skinned, and cut up the meat of a caribou ready for loading on a sled in 25 minutes, at 30 degrees below zero. The difference was entirely due to knowing what to do, and making it snappy.

MAMMALS REQUIRING SPECIAL TREATMENT

Bears

Bears necessitate more hard manual labour than most species, as they are generally fat and in any case the skin adheres tightly to the flesh and must be cut away. Scraping the grease from the skin is a long and tedious operation which should be done as soon as possible, as the skin will dry rapidly in spots where the knife cuts have removed the fat, and it is very easy to gash the skin at these points, or to shave off a patch of the hair roots so that the hair will drop out. The skin of a polar bear is most readily cleaned of fat after it has been spread out on the ice or snow and frozen flat and smooth. By a fortunate provision of Providence, polar bears and Eskimo are frequently found on the same range, and an Eskimo woman with a curved-edged *ulun* or woman's knife will shave the blubber from a bear skin with high professional skill and startling rapidity. It will pay any collector to watch this operation once for his own education. The same results may be obtained by using a strongly curved skinning-knife with bevel-edge and stretching the skin across the thigh or over a rounded beam. With a little practice, cultivating a sweeping, outward, shaving stroke, with the back of the hand up, regulating the pressure by the "feel" of the blade on the inside of the skin, the blubber may be loosened and rolled up very rapidly. The removal of the blubber is an absolute necessity, whether the skin is to be salted or dried.

The bear's feet should be skinned to the base of the claws, and the thick, hairless pads on the soles should be left intact and attached to the skin. The foot is most readily skinned by continuing the leg incision around the inner side of the foot at the edge of the hair. The ordinary hunter or trapper, white man or native, usually cuts off these pads, leaving them attached to the carcass, and most "hunter's skins" come into the market in this shape. Though this mutilation does not deduct from the value of the skin for a rug, it does deduct from the scientific value. Bears, or any other mammals with thinly haired under parts, should have the opening cut made along the middle of the back, if the specimen is to be mounted standing erect on the hind feet.

If Indian or Eskimo assistants are employed, they must be watched very carefully, as many of them have traditional methods of their own for cutting and removing skins of most animals, to say nothing of superstitious taboos, one of the most annoying being the common practice of leaving some part or tuft of the skin attached to the skinned carcass. The feet, and the perineal and genital regions are most subject to such mutilations, and the collector who would bring out a perfect, complete skin must be continually on the job.

Seals and Walruses

Seals have the skin lined with a coat of soft, greasy blubber, and are most readily skinned by cutting a deep gash along the under surface, clear through skin and blubber, and dissecting and rolling the carcass out of the skin. If the skin is to be mounted, it is well not to bring the opening cut farther forward than the breast and to make lateral cuts to each flipper. The blubber may be cut away in a mass with a curved knife as in the case of a bear skin. By holding the skin taut with one hand, one may usually avoid cutting the skin—with practice. If any seal-hunting natives are around, it will usually be cheaper and easier to have this rough cleaning done by a professional, although the finer points of the head and flippers had better be done by the collector himself. Bearded seals and sea-lions should have the lips carefully skinned and thinned down around the roots of the "whiskers" so that the salt can strike in; otherwise the bristles are apt to slip out.

The ordinary method of preserving seal skins has been by salting, but as it has been found that brine changes the colour of hair seals very much, the skins should be dry-cured if possible, or at any rate, partly dried, and the salt kept on the flesh side away from the hair (*See* page 74).

Walrus skins are preferably thinned down at once with the curved skinning-knife, to not more than one-fourth of the original thickness, and the skins salted in bulk. Dead seals and walruses designed for scientific specimens should not be exposed to sunshine for any length of time before skinning, as the cuticle will blister in a remarkably short time.

Beaver

The beaver is handled as an open skin, with opening cut along the median line extending to near the tip of the tail on the lower side. The broad, flat, scaly tail presents no particular difficulties, but care should be taken not to cut gashes through the skin as there is no hairy covering to conceal stitches. The feet are skinned down to the toe-nails and the webs between the toes split or separated in the process. To do this, it is necessary to split the sole of the foot from a point near the junction of the middle toe to the centre of the heel in the hind foot, and to a short distance beyond the wrist in the fore limbs. As the skin is rather heavy and fatty, it is rather hard to dry properly without undue stretching, and is best dry-salted and later tanned in the laboratory.

Muskrat

The muskrat is skinned and made up like an ordinary small mammal. The only abnormal feature is the tail, which is long, scaly, sparsely haired, and laterally constricted, so that its height is much greater than its width. The tail must be split on the under side nearly or quite to the end in skinning. To get a thin tail filler the best method is to whittle a tail of soft wood (preferably white cedar) to the same size and shape as the skinned tail, and long enough to extend about halfway through the body. The same high, narrow effect may be obtained by wrapping two wires tightly with cotton and then wrapping the two together with fine fibre cotton. The latter method is used in mounting a muskrat if the tail has to be bent.

Porcupine

The Canada porcupine is a formidable-appearing animal and may be embarrassing to a novice, as the quills are rather loose and the barbed points are painful weapons. A porcupine killed with a club is apt to have the skull broken into fragments or to have many of the dorsal quills flattened or knocked off. The best method of killing is by a small rifle bullet through the body, or by shotgun through the thin-skinned, unprotected under parts, which avoids riddling and tearing the quills. The selection of a shot is a simple matter, as the porcupine will generally climb a few yards up a tree and stay there. The porcupine has no quills on the under part of the body and throat and the skin may be rapidly removed without injury from a single quill.

Make a cut along the median line from the throat to the tip of tail, and cut the skin from the body with long, sweeping strokes, turning the edges of the skin outwards and rolling the skin back with the quills inside. As the roots of the quills are deeply embedded in the skin, be careful not to cut off their bases. The tail is very thick and muscular, with the skin grown tightly to the flesh, so it must literally be carved out of the skin, avoiding if possible cutting any gashes in the tail skin, particularly on the dorsal side. As the skin is usually fat, it needs considerable subsequent cleaning with knife, shears, and scraper. It is generally best to salt a porcupine's skin in the field, and defer cleaning until the skin reaches the laboratory, and has hardened a bit. Although the porcupine is rather large it is usually easier to handle in the museum as a made up "study skin." In making up the skin, stuff it rather smaller than natural size and flatten the body considerably, not more than 3 or 4 inches deep, otherwise the specimen will take up too much case room and be a problem generally. A porcupine skin can be handled with less danger from quills if a narrow piece of wood is used to stiffen the body, as in the "California style" of rabbit skin (page 80), letting the stick project far enough behind the body for the legs to be tied to the stick. The projecting stick may be used as a handle in lifting the specimen.

Hares and Rabbits

Although these animals are rather large and bulky, they are best made up as study skins. The skin is thin and tender and brittle when dry, so that when flat or cased the heavy legs and feet are apt to be torn off. The skin is removed in the manner usual for small mammals.

As the head is angular and irregular in shape, with broad cheeks, narrow nose, and depressed muzzle, care should be taken to make the head filling as near the natural size and shape as possible. Crumple up a hard wad of paper for a core and wind cotton, tow, or oakum around it as tightly as possible, to make a firm artificial head. Leave the leg bones attached to the feet and wrap them loosely with cotton or tow. If the leg bone is broken make a rough splice by thrusting a bit of stick or wire into each of the broken ends. Cut two pieces of fairly stiff wire (iron or brass, annealed, No. 16 or 17), sharpen one end of each, and thrust them into the heels from the outside, along the hind leg bones and into the fore leg on each side, and out at the wrist. Arrange the skin in proper length and tie the leg bones fast to the wires, which run parallel to each other through the body (Figure 25). Make an artificial body of soft filler, thrust the anterior end into the back of the head filler and spread the posterior part around the dorsal side of the connecting leg wires. Put in flat bunches of cotton where needed to fill out around shoulders, hips, and base of tail. Also place a layer of cotton along abdomen to cover the wires where exposed. The tail should be supported by a small wrapped wire. If wires of the proper size are not available, the fore and hind legs may be connected by slender strips of wood lashed to the leg bones, or the legs may be fastened to a stiff stick running the full length of the body. The essential point is that the legs must be fastened in place and the body connected rigidly fore-and-aft or the dried skin will become easily broken in handling.

Arrange the body on a board for drying, pinning it down with heavy pins or slender wire nails. The cartilage should have been skinned out of the ears as far as possible, at least loosened from the skin on the outer side of the ear. The base of the ear may be stiffened with a curved piece of cardboard or stiff paper. After the body is pinned down the ears

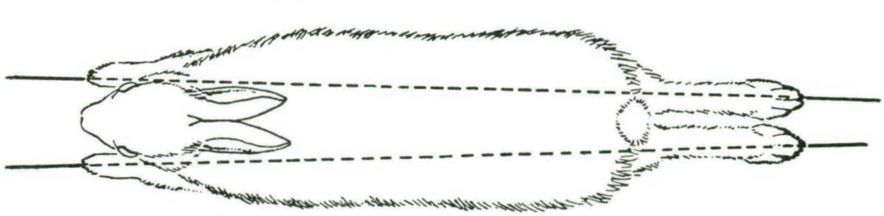

Figure 25. Method of wiring a rabbit skin.

should be laid back on the head and neck, parallel with each other, and a firm, pointed roll of cotton thrust down into the external auricular opening of each to keep them in shape until the skin has dried. The cotton roll may be pulled out after the skin has dried. If the specimen has to be moved before it is thoroughly dry, it is well to pin down each ear and to fasten the ears together by a stitch through their inner edges.

"The California Style of rabbit skin." Although the method described above makes a good exhibition specimen skin, the technique involves rather too much taxidermic skill, and in a large series the specimens take up too much room. The National Museum of Canada has lately adopted the "California style" as developed by Mr. Adrey A. Borell. Some of the advantages are: flatness, giving economy in storage space; firmness of the fore and hind limbs, thus preventing breakage in transportation and later handling; and simplicity of technique in the field, doing away with the necessity for pinning while drying. As described by Dr. E. Raymond Hall, the "stuffing" of the body is prepared by cutting out a piece of corrugated cardboard outlining the body and attaching a stiff wooden stick to the cardboard by means of thread or wire at two places. The stick projects forward to the head and far enough posteriorly to allow of securing the hind feet to this central support. A thin layer of cotton is laid over this "paddle", which is inserted in the skin or the skin turned back over it. The posterior square end of the cardboard is then cut to fit the skin, and wire or, preferably, slivers of wood or sticks are inserted, one in each leg. The skin is sewed up, and the hind legs are secured to the median stick by a stitch of strong thread or preferably a bit of thin wire passing

through inner side of each foot and fastened to stick by twisting ends of wire together with pliers. The fore legs are laid out flat along each side of throat and secured to the body by a hidden stitch between the toes. The ears are spread out with dorsal side down, and held together by a stitch through inner edge of both ears.

Flying Squirrels

Flying squirrels have broad, lateral folds of skin extending from the wrists to the ankles and covered with fur on both upper and lower surfaces. These double folds of skin should be separated and the inner side of the skin treated with preservative. In filling the skin do not let the body stuffing spread out into these lateral folds and make a sofa pillow effect. The body filling should be firmly wrapped and the legs should be held in to the body by thread tied around the front of hips and back of shoulders. Lay out the skin as in the case of an ordinary small mammal. Spread the lateral folds out smoothly and if they show a tendency to spring up, cut a curved piece of pasteboard to cover each side and pin these down close to the body until the skin is dry.

Bats

It may be well to spread fully the wings of one or two specimens of each species and pin them out on the drying board. Spread bats' wings are easily broken and for that reason most collectors make up the bulk of their bat skins with the wings neatly folded at the sides (Figure 21). However, in many species of bats important specific characters are derived from comparative measurements of length of each finger, and length of metacarpal bones, and it is often difficult to measure these parts in a closely folded, dry skin. The writer has frequently found it necessary to relax such a skin before the desired measurements could be made. For this reason, he has recently been advocating that bat skins (particularly of the genus *Myotis,* the little brown bat and its near relatives) should be made up with the wings partly spread, with each finger pinned out far enough that the different joints may be examined and measured separately (Figure 26). In addition to the three usual measurements (length, tail, and hind foot), bats should have additional measurements of the ear: height of ear and height of tragus (*See* Measurements, page 44). The size and shape of the ear form important specific characters in some species, and the collector should try to have the ears dry in approximately natural state, keeping them in shape by occasional pinching into shape while the ears are still partly flexible. Major Allan Brooks stiffens the ears of the longer-eared species by painting the *inside only* with shellac; if care is used the tragus will stand up. It is also advisable to make an outline sketch of the ear, including tragus, on the back of the label.

Skunks

Most collectors are shy of handling skunks, but when killed properly and cleanly, a skunk is no more difficult to skin than a weasel, mink, badger, wolverine, or any of the other Mustelidae, all of which have scent-glands secreting a more or less disagreeable musky odour. The skunk is much less dangerous than is generally supposed. It never throws its scent except in self defence, and then not more than 8 or 10 feet, although the spray may be carried somewhat farther in a strong wind. Various experts state that the surest way to kill a skunk without smell is to drown it, but the writer can guarantee no safe way to get the skunk

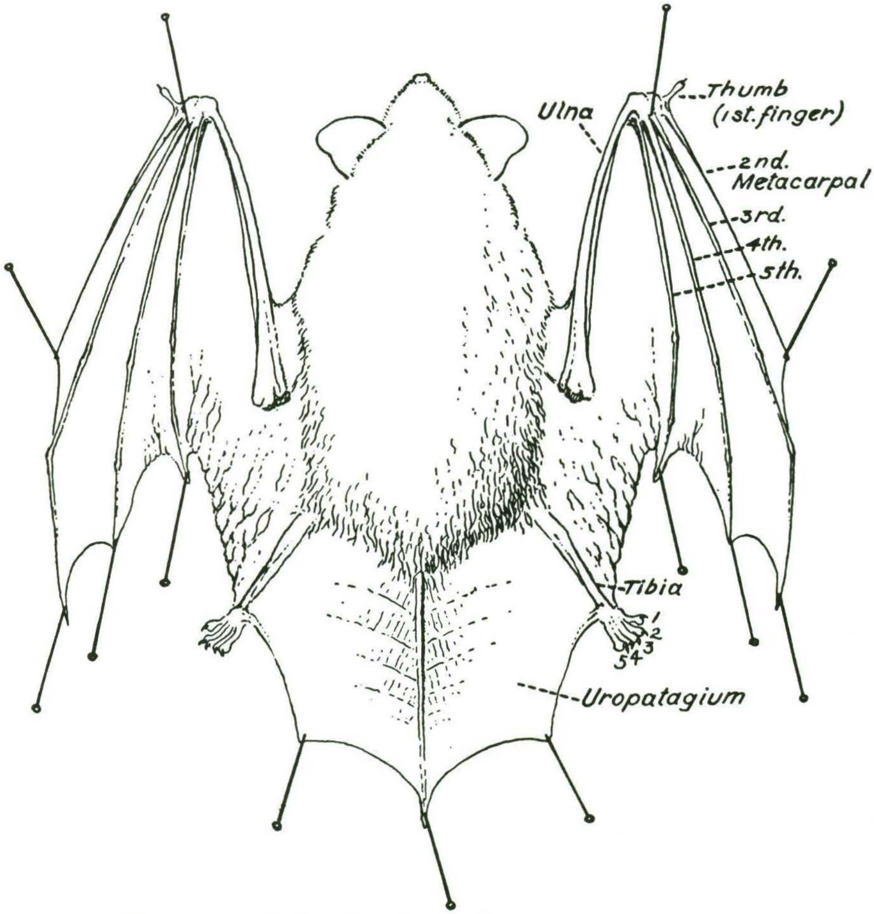

Figure 26. Drying bat skin with wings partly spread.

to water. Seton (1929) and other authorities assert that the skunk is harmless if held up by the tail, but like other methods of "belling the cat" it is not always safe for the amateur. The skunk, however, is a careful and cleanly animal and does not willingly soil its own fur, even if caught in a trap. It may be easily and painlessly killed by approaching cautiously from one side and dealing a heavy blow with a club across the small of the back, over the kidneys. Shooting spoils the skin for the fur market, and if the animal is not instantly killed some fluid may be ejected. However, a charge of small shot at medium range in the lower part of the back will not spoil the skin for a specimen, and will usually cut the spinal cord or paralyze the hind quarters so that no scent will be ejected. Any faint skunk odour will disappear from the skin in a few days, but if badly saturated the skin should be scraped and washed in gasoline, which removes the grease from the skin at the same time. A mild solution of vinegar ($\frac{1}{2}$ cup to 1 quart of water) is helpful in removing skunk odour from skins, clothing, and dogs.

PELTING SKINS (FOR THE FUR TRADE)

Cased Skins

All skins of weasel, ermine, mink, marten, otter, muskrat, opossum, and skunk should be cased and dried on stretchers with the *pelt side out* and the *fur inside*. Fur buyers and game officials usually judge whether a skin is "prime," that is, taken in open season, by looking at the flesh side of the skin. If "prime" and the hair fully grown, the flesh side of the skin is a light creamy, whitish, or parchment colour, while "unprime" skins, taken out of season, show dark reddish, brownish, or blackish areas over the whole or part of the skin, due to blood and pigment cells at the roots of the new growing hair. The details of removing the skin for casing have been described on page 67.

Skins of the fox, wild cat, lynx, and wolf are also cased and dried on stretchers (Figure 11), but before the skin has become too hard and stiff it should be turned so that the fur is on the outside. Sometimes part of the skin will need to be softened slightly with wet fingers or a damp cloth and worked and twisted a bit before it can be turned right side out. The skin should be hung up for a while to dry this superficial dampness before packing it away. Many northern trappers improve the appearance of their fox furs by suspending them by the head from a strong line at a safe height from the ground for a month or so during March, April, and May. The strong winds drive snow and sleet particles into the fur and remove particles of blood and dirt, and the frequent periods of bright sunshine and snow glare have a cleansing and bleaching effect. The trapper should be careful to split the tail sheath of all skins that are hung up exposed to snow, as the snow in melting may run down into the sheath and cause the hair to fall out. Any furs that have become mussed by baling, packing in bags, or otherwise, may usually be freshened and fluffed out by hanging in the wind for a few hours.

Some fur trappers mistakenly overstretch the skin of a fur animal trying to make a large skin out of a medium or small one. This trick does more damage than good as an overstretched skin becomes thin and the fur less dense, causing reduction in sale price. On the other hand, if the skin is allowed to wrinkle, it will not dry properly and the hair is apt to slip off in the creases.

Open Skins, or Flat Skins

Bear, beaver, raccoon, wolverine, and seal should have the skins removed by opening so that the entire skin may be stretched flat on a board or on the ground.

To prepare an open pelt, cut the skin down the abdomen, from the lower lip to the tip of the tail, and from the feet cut up the back of the hind legs, and on the inside of the fore legs to join the median cut. In cold winter climates the skin may be spread out on the snow and frozen flat and smooth, after which it may be hung up outdoors. In the dry, cold atmosphere of northern Canada, the moisture will evaporate rapidly from a frozen skin hung up exposed to the wind. In warmer regions the skin may be tacked or pegged out, or the corners held down by weights to dry.

Beaver skins for the fur trade have the tail and feet trimmed off and the skin stretched in circular shape on a wooden hoop, the skin laced to the hoop by a thong passed through holes cut in the margin of the skin. The tail and feet of muskrats, and the flippers of seals, are also cut off before stretching. Such skins, however, are worthless for scientific purposes, except for showing condition of the pelt at the time of taking.

Most natural history museums do not have adequate collections of the valuable fur-bearing mammals. Competent trappers are not often available, winter field work is difficult and expensive in the haunts of the rarer mammals, and high prices for fur prevent extensive purchases. The skulls of such mammals from different regions are of great interest to the zoologist for study of geographical races, and to the palæontologist for comparison with fossil forms. Skulls of skinned mammals are a mere by-product of the trapping business, and are generally thrown away, but the National Museum will be grateful to any trapper who will take the trouble to donate skulls with data.

CHAPTER IV

COLLECTING AND SKINNING BIRDS

COLLECTING BIRDS

Mounted Birds and Bird Skins

The general principles of bird collecting are the same as of mammal collecting, and as practically the same tools, preservatives, and equipment are required, the prospective bird collector is referred to the preceding pages of this bulletin.

By the general public, a bird specimen is usually thought of as a bird stuffed, mounted, or "set up" in a more or less natural or life-like attitude for exhibition as a trophy, ornament, or a scientific or educational exhibit. When a professional taxidermist is available, the amateur who merely wishes to preserve a trophy will generally find it best to send the bird "in the flesh" in as clean and fresh a condition as possible.

Mounted birds are difficult and expensive to prepare, bulky to store, easily broken or damaged, and the colours are invariably subject to fading in daylight. In scientific museums and large private collections, scarcely a fraction of one per cent of the bird specimens are mounted, and nearly all such specimens were originally collected as "bird skins"—also known as "scientific skins" or "study skins." The "scientific skin," so-called, is the skin of the bird, removed, cleaned, treated with preservative, with the skull left inside attached to the head skin, and enough loose filling placed inside the skin to make it resemble the dead bird. The skin is laid out to exhibit in the best manner the external features of the bird, the colours of its plumage, and the general form of bill, wings, and feet.

Bird skins may be mounted at any later time, and the general shape or "make" of such skins is not important. But as by far the greater number of skins are kept for purposes of study, it is important, whenever reasonably possible, to fill and lay out the skin in a smooth, conventionally uniform position for convenience in comparing series of specimens. Any student who has occasion to compare large series of birds and mammals prepared by different collectors with various methods knows how difficult it is to make good comparisons of colour patterns and texture when the feathers and fur are dried in a careless, rumpled and unnatural state. Some of this waste of time and material is due to careless and inexperienced field collectors, but much is also due to the urgent desire for quantity rather than quality by market-hunting professionals and expeditions that are avowedly out to be first in any imperfectly explored area, and get as many specimens as can be picked up and possibly named as new. Dr. Witmer Stone, for many years curator and director in the Academy of Natural Science of Philadelphia, which is noted for the quality of its scientific material, wrote in 1933 (*The Auk,* 50:2, 243), "There is no excuse in these days for the carelessly made specimens which have hindered research in the past and made our museum collections unsightly."

For mounting purposes, good typical specimens of both sexes in full fresh plumage are generally selected, but the scientific or research collection needs specimens of both sexes at different seasons—in spring nuptial or breeding plumage, winter plumage, moulting or transitional plumages—and the young at various stages from the downy young stage to full adult plumage stage. With some birds, such as the large gulls, there may be different stages of plumage every year up to the fourth year. It follows that whereas many a skin is not suitable for mounting, it may have important scientific value if its history is recorded on the label.

Many species of birds may be captured alive in the various kinds of traps used by bird banders (Lincoln and Baldwin, 1929). Such specimens have the advantage of being clean, and the collector has the advantage of being able to carefully select the specimens he wishes to preserve. One feature which might be noted is that when "birds of the year" are banded, the collector may have a chance to examine adult birds of known age in succeeding years, by studying the "repeats" or birds that are trapped more than once. The bird collector has one great advantage over the collector of small mammals, in that he can pick up his specimens on casual 1-day excursions, shooting the specimens he desires as he sees them, whereas the mammal collector is generally obliged to set traps and visit them again the next day.

Superficially, the collecting and preserving of mammal skins may appear to be easier than working with birds. The skin of the mammal is usually tougher and the hair well set and durable, so that it may be roughly handled and put through various cleaning processes with impunity. In practice, however, a bird is generally more easily and rapidly skinned than a mammal of corresponding size, and on the average more birds can be put up in a day with less trouble.

Collecting Birds in the Field

A large part of the trouble with bird skins may be prevented by proper handling at the start. The feathers should be kept unruffled and as clean as possible, and the skin untorn. If a small bird is caught alive, it may be quickly and cleanly killed by pinching the bird sharply under the wings with the thumb and forefinger. The bird has a high temperature, rapid circulation and respiration, and compressing the ribs over the heart and lungs results in almost instant death. With birds larger than small hawks this method will not work well as the fingers may not be strong enough to give the needed pressure. Bearing down with the knee, assisted by the weight of the body, on the side of the bird's chest, will produce the same effect. Birds have hollow bones, and these and other cavities and air sacs are connected with the lungs, so that a bird may suck in enough air through a broken bone to prolong life for some time. In such cases, the bird may be instantly killed by opening its mouth and thrusting a penknife blade into the base of the brain, the method used by dealers in killing poultry. Care should be taken to make the incision far back, as the bird's brain is situated well behind the eyes. A plug of cotton should immediately be placed in the mouth to absorb any blood flowing. The bird collector should always carry in his pockets, a knife, a pair of fine-point forceps, a bunch of cotton batting, and a bottle or can of dry absorbent powder.

In shooting birds, be careful to use shot of proper size. If the shot is too large it will make larger holes than necessary. The smallest sizes of shot (No. 12 and "dust shot") will usually make such small holes that little blood will flow, but even the smallest shot will break legs, riddle beaks, and split feathers if used in large charges at close range. The collector should carry shells loaded with different sizes of shot, and carry these in different pockets so that the proper kind may be found when wanted. Game birds will usually be shot by ordinary shooting methods, and as they are usually obtainable in quantity, sportsmen are apt to use heavy charges to make large scores. Most of the species collected are non-game and will have to be stalked and "potted" in any way that will allow the bird to be killed at just the right distance to make a clean, perfect specimen. The collector of scientific material is not killing for "sport," and rare or useful species should not be sacrificed to make a sporting shot. As some birds, such as warblers, are very restless and active, shooting them in thick bush is often as difficult as wing shooting, and "squib" loads of dust shot may have to be used in the "Aux." barrel at very close range (*See* Coues, 1903, i, page 158).

Methods vary for finding the different species. Some of the shy, meadow-and-marsh haunting species are more readily flushed by noisily tramping through the grass or water, as some of these birds sit close and will scarcely rise until nearly stepped on. In thick brush, the chances are that any bird or mammal will see or hear an intruder long before the latter suspects, and if afraid will flit away. In such cases it is best to walk slowly and carefully, and avoid making unnecessary and alarming noises. It is also advisable to sit down frequently, in places where there are small openings in the woods. Most of the small birds are curious and will come back to investigate. They appear to pay little attention to shape or colour of a large object, but notice the slightest movement. If the observer can remain without moving a muscle he may very soon have a number of birds around him. Making a squeaking noise by sucking with the lips pressed against the back of the hand, or merely squeaking with the lips will often bring numbers of birds very close, particularly during the spring and autumn migrations. Close imitation of the note of any particular bird may help. Various duck calls have some effect on water birds, and the writer has seen an Eskimo call back a flock of whistling swans, which has passed half a mile beyond, by a skilful imitation of the swans' notes. With the small woodland birds, the lure generally appears to be curiosity about any unusual bird-like sound, or the sound of a bird in distress. In British Columbia it is said that no bird collector can be really successful without imitating the note of the pygmy owl, as most of the small birds in the vicinity will rally at the sound of their favourite enemy. Other observers have drawn small birds around by setting a mounted screech owl in an exposed place and letting nature take its course.

Preliminary Treatment of the Fresh Bird

The freshly killed bird should be handled carefully, as two or three minutes' attention at the start may easily save an hour's time later. Carefully scrape any drops of blood from the feathers with a knife blade. A small wad of cotton dampened with water or saliva may be used to wipe off blood stains, but acetone, or a mixture of acetone and carbon tetrachloride is better. A small bottle of this carried in the pocket while collecting will be a great help. The bird collector should carry a pair of fine-pointed forceps in his pocket or game bag, the tips shielded by sticking into a small cork. A pin or bit of wire will do for probing in an emergency. If there are any visible shot-holes, they may be plugged with a bit of cotton, and fine cornmeal, whiting, sawdust, or dry dust sprinkled around the surrounding feathers. Shove a large plug of cotton into the mouth and throat to keep blood and digestive fluids from running out. If blood oozes from the nostrils place a strip of cotton over them and across the forehead, stuffing the ends on each side into the throat, and plugging them with another wad in the throat. Avoid plugging soft nostrils as the form may be changed. If either of the eyeballs has been punctured by a shot, shown by water running out or by deflated appearance, insert the point of forceps and pluck the eyeball out, sticking in a wad of cotton to take its place. If a broken eyeball is not removed, the sticky liquids are sure to run out on the feathers of the head and neck, and nothing is harder to clean off.

In very hot climates, where small birds may decompose and lose feathers on the throat and abdomen in a few hours, it is well to carry a hypodermic syringe and inject a preservative into the throat and abdomen. Weak formalin (1 part to 25 of water), or a saturated solution of alum, or a weak carbolic acid solution may be used. The abdomen is injected through the vent, and the throat has a little of the solution squirted into the gullet.

A single specimen may be carried home by the bill or feet. The best method is to make a funnel or cornucopia of fairly stiff paper, such as the glazed or sized paper used in some of the larger magazines. A few loose sheets may be carried and rolled and pinned as needed, but the writer has found it more convenient to roll a few funnels of different sizes and paste them to retain proper size and shape. The different sizes may be telescoped and carried in the bag, and may be used again and again. They retain the specimen in better shape than extemporized wrapping. After cleaning and plugging the bird, drop it head foremost into the paper funnel, and fold the edges in, securing the edges by a pin if necessary. The specimens may then be placed in a basket or other receptacle in which they will not be squeezed or the tails mussed. If wing or tail feathers are broken, they are almost impossible to repair, but if only bent or twisted they will straighten immediately when held in a jet of steam from a kettle. The ordinary hunting coat pocket is not very good for carrying bird specimens, as they are apt to be shaken up or smashed in the excitement of hunting. A stiff haversack, or best of all, a fishermen's basket creel, is very useful, and may be readily slipped off when necessary.

Measuring Fresh Birds

The old collectors were accustomed to take three or four measurements: length (from tip of bill to end of longest tail feather); wing (in straight line from bend of closed wing to tip of longest primary quill); tail (length of longest tail feathers, measured from the bony base of the tail); and bill (length of culmen; tip of bill from edge of frontal feathers to tip of upper mandible). As all of these measurements, except the total length, may be readily taken from the dried skin, there is no particular advantage in taking them from the fresh bird. As most research workers prefer to make their own measurements when studying the specimens later, many collectors have dropped the habit of taking any bird measurements in the field.

As the " Length " of a bird is frequently given in popular books for identification, it is well to record on the label the length of the fresh bird. The " Extent " (E.), or spread of wings from tip to tip, is also of interest, particularly in large birds, and is taken by holding the tip of one wing down on the floor or the edge of a table and pulling the other wing out to its fullest extent and marking the distance. Chapin (1929) recommends taking also the "Length of body"—the distance measured with the dividers from the anterior surface of the shoulder to the vent, or if the bird is skinned, to the tip of the pubic bones. The length of neck can be derived when the total length, length of body, and the length of tail are known, and the measurements are very useful if the skin is ever mounted.

Colour Records

Collectors have always been advised to keep in their notebooks a record of colours of iris, bill, feet, and any soft or bare parts of birds, as these parts dry out and lose their colours, entirely or in part, shortly after death. Some of these colours differ with the sex, and change radically with age and season. Some collectors do keep records of such things, but many young collectors mistakenly assume that these matters are all known and recorded by the pundits of ornithology, and fail to make original records. If it happens that all the seasonal colours of a bird are already well known to ornithologists, the colour tints may give a clue to the age or sex of a specimen when it can not be determined from the plumage.

Many birds are so rare that only a few specimens have been taken by professional ornithologists, and perhaps only in certain stages. As the systematist, taxidermist, and bird artist wish to know the colouring of any particular specimen, Major Allan Brooks and others have long been urging collectors to record the colours of soft parts of birds *on the label.* Every zoologist should be to some extent familiar with colour names, and if possible own a copy of Ridgway's "Colour Standards" (1912), or have access to a library copy. Failing this, frequent reading of bird descriptions in standard works and comparing of these with specimens will give a general idea of the more common shades of colour. At the very least, he can record whether the eyes of a young bird are " brown " or " yellow ", or its feet " yellowish " or " blackish." If the collector has any skill with water colours, it is a fine thing to make colour sketches of bill or feet, or a rough diagram with coloured pencils.

SKINNING BIRDS

When skinning birds or small mammals, place a sheet of newspaper on the table and change it for another when it becomes soiled. Immediate notes may be jotted on the corner and at the finish the offal and loose paper may be rolled up and burned or buried. On long trips away from civilization a piece of oilcloth is handy to skin specimens on, and keeps tools from getting lost.

It is best to let a bird lie for a short time after death for the blood to coagulate. A freshly killed bird bleeds freely and it is well to keep blood away from the feathers. Plug the throat with a fresh wad of dry cotton and put a fresh pad on the nostrils if the blood continues to ooze out. If the body has begun to stiffen, the wings will be in the way, and the humeri (upper bones of the wing) may be snapped in two as close to the body as possible. This may be done with the thumb and fingers in small birds or by a tap with a knife handle or hammer in large birds. If a large bird, such as an eagle, hawk, or owl, is to be mounted with spread wings, it is just as well to leave the humerus intact inside the skin.

Lay the bird on its back and part the feathers in a straight line along the middle of breast and abdomen. The feathers of birds are arranged in well-defined patches (pterylæ), and in most land birds the parting will show a more or less bare strip along the median line. In water birds this part may be covered with short, downy, under feathers. Make an opening cut from about the middle of the breast bone backward to the vent. To avoid cutting through the wall of the abdomen, a small opening may be cut on the breast and the handle of the scalpel thrust underneath the skin to work it loose at sides and backwards. Slipping the edge of the scalpel under the loosened skin and cutting upwards, the cut is extended to the vent. If the abdominal wall is opened by a shot or knife cut so that the intestines or juices come out, there is no great harm done, but the hole should be plugged with cotton. From the time the first cut is made pinches of cornmeal or fine sawdust should be sprinkled freely upon the flesh to absorb any fluids and to dry the surface so that the feathers will not stick to it.

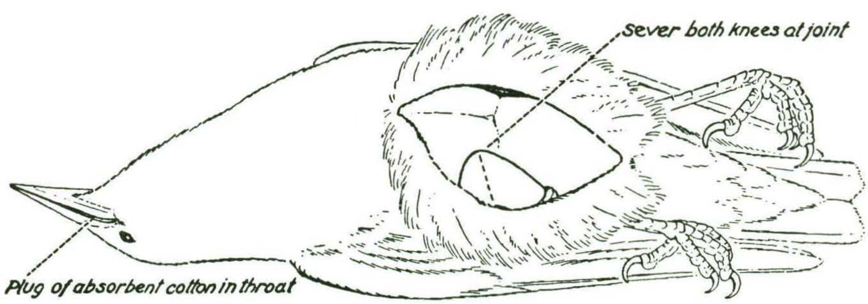

Figure 27. Opening cut for skinning a bird.

Loosen the skin along the sides of the body and the knee joint soon becomes visible (Figure 27). Take hold of one of the feet from the outside and push the knee farther up into view. The leg should be severed at the knee joint, in small birds by clipping it in two with the scissors, and in large birds by cutting across the tendons with a knife and then separating the bones. Some collectors leave skinning of the leg until later, but the better practice is to skin the leg at once to prevent the weight from tearing the skin. Turn the skin of the leg wrong side out, working it loose from the flesh with the thumb-nail, as far as there are any feathers. If the tibia is partly scaled, as in sandpipers, the leg can be skinned only part way down, but if the tarsus is feathered, as in some hawks and owls, it may be skinned to the toes.

In skinning large birds with touch skins, the skin may be loosened along the sides and lower back with fingers and handle of scalpel until the skin is free around the rump. Then with the scissors first cut through the lower end of the intestine close to the vent, and then through the base of the tail far enough forward to avoid cutting the basal ends of the rectrices (tail quills). Never cut off the ends of .the tail feathers or they will fall out. If any of the tail feathers come out they may be fastened in before filling the skin, by running a small wire into the base of each feather from the inside; or, in a small bird, by sticking it in later with glue.

With small birds, or large birds with tender skins and loose feathers, or with skin growing tightly to the hip bones, it is almost impossible to work the skin loose all the way around before cutting off the tail. The skin is almost sure to rip down the side just behind the legs and the loose feathers on the rump are apt to drop off in amazing quantities. The skin is carefully loosened back of the legs sufficiently to allow cutting the lower

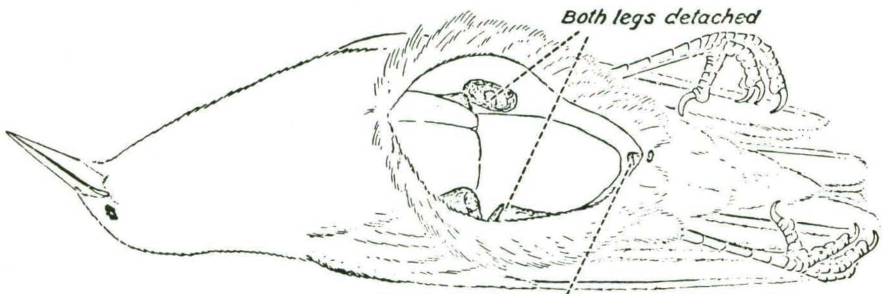

Figure 28. Severing tail from body of a bird.

tip of the intestine. Absorbent material is dusted in and if necessary a plug of cotton put into the out. The tip of the pointed scissors is then carefully inserted just underneath the tip of the posterior vertebræ just ahead of the tail bone and the tail bone is snipped off (Figure 28). Use great care not to punch a hole in the skin of the rump. Bend the tail back on the body leaving the muscles attached to the tail for the time being and stripping the ends of the muscles off the lower back. Continue to keep all wet surfaces covered with absorbent.

Now hold the body up by the hips, and with the thumb and fingers of the other hand, and the scalpel if necessary, separate the skin of rump, back, and sides from the body. Keep dusting on absorbent from time to time, and by manœuvring the hands and fingers try to keep the loose ends of the feathers away from the flesh as the work goes on. Loose fluffs of cotton laid along the dividing line between the skin and flesh will also help to keep the feathers away.

After getting the tail out of the way, the beginner will find it easier to hang the specimen up by one or two cords with hooks stuck into the pelvis. This enables him to use both hands in the actual skinning operations. After a little practice the skinning operations will become familiar and the hooks will not be needed, although it is often convenient to suspend a very large bird by a slipknot around the hips just anterior to the stumps of the thighs. In skinning a suspended bird, be careful of blood spurting over the plumage from severed veins. Dripping blood may be avoided to some extent by tying cords tightly around the base of wings and upper part

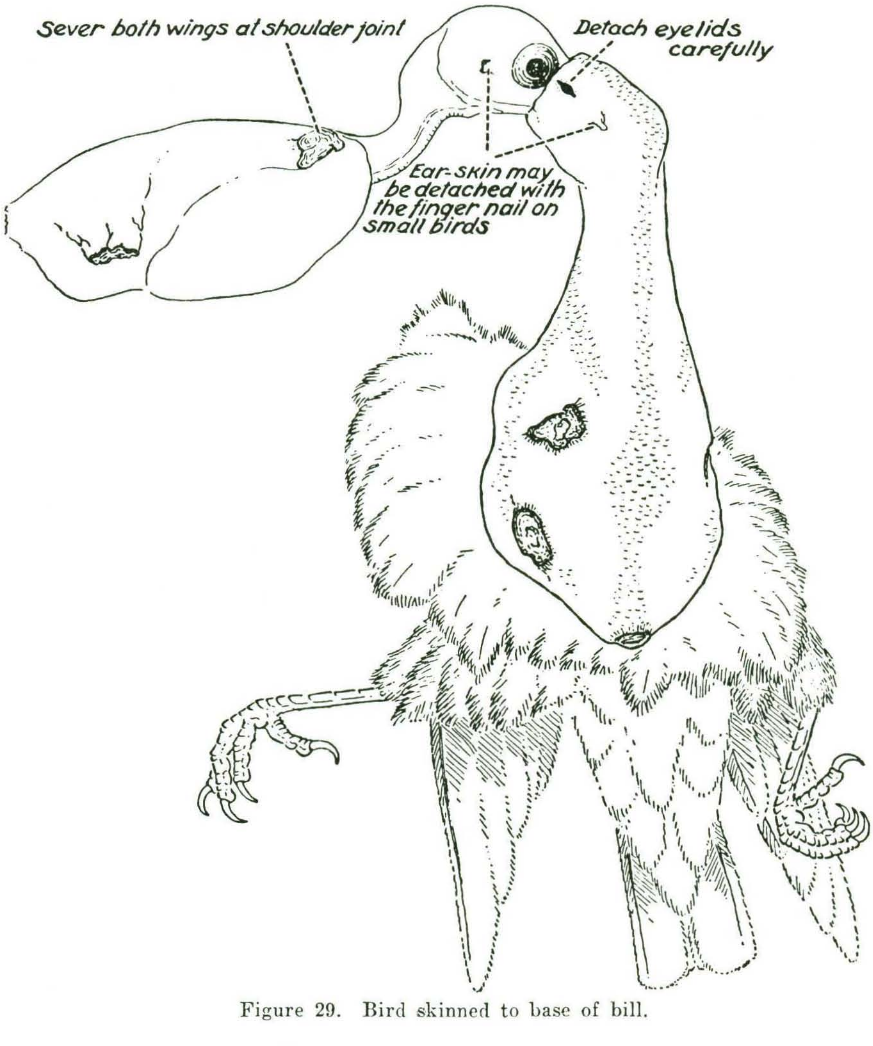

Figure 29. Bird skinned to base of bill.

of neck before cutting off wings or working on the head. With small birds any blood flowing may be absorbed with cotton or dry absorbent. When the bases of the wings appear they should be cut off near the shoulder joint, and the skin turned back over the neck until the head is reached (Figure 29).

The skin should be worked carefully over the head, being pushed from the back of the head with the thumb- and finger-nails. Do not try to pull the skin over the head all at once, but work it slowly from all points, gradually stretching the skin until it slips over the widest part of the skull. The amount of strain that the skin will stand without breaking, both in skinning and in slipping back over the skull, can only be learned by practice. If the head skin splits along the side during the operation it is hard to mend without leaving a trace, but a few stitches with fine needle and thread during the stuffing operations may possibly repair most of the damage. As the back of the head comes through, there is seen on each side the membranous tube of skin running into the aperture of the ear. These bits of ear skin may be pulled from their hollows by grasping them between the nails of thumb and forefinger, or with the forceps. In some large birds it is necessary to cut the skin of the ears as close to the skull as possible to avoid leaving an opening visible on the outside.

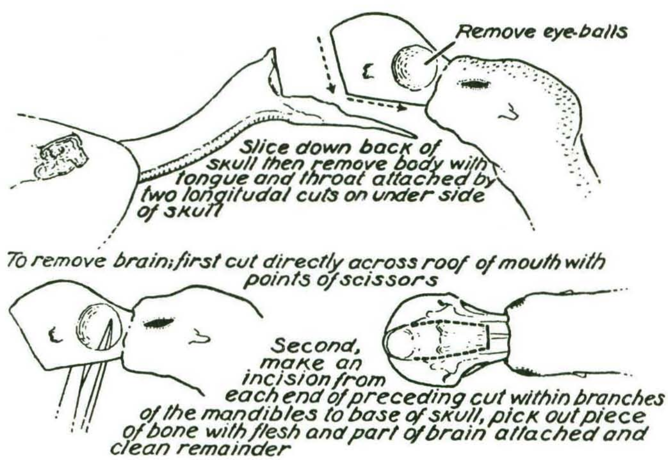

Figure 30. Detaching body and cleaning skull.

A little farther on the eyeballs appear under their transparent membrane which connects the eyelids with the eyeball. Cut through this membrane carefully but be sure not to cut the eyelid. A cut eyelid can never be properly mended. The loose membrane should be carefully trimmed away from the eyelid or it will be in the way when the cotton filling is put in the eyeball. Continue skinning to the base of the bill, where the feathers stop, both above and below (Figure 29).

Pry the eyeballs out of their sockets with the handle of a scalpel or with the large forceps, but avoid breaking the eyeballs, as the juice is almost sure to run out through the eyelids and gum up the feathers of the head and neck. Owls and other birds which have eyeballs stiffened with bony plates should have the eyeballs left in place, but they should be punctured and drained (*See* page 110). The tongue and floor of the throat may be pulled away with the forceps, cutting where necessary in large birds (Figure 30).

As the skin of the head merges into the skin or horny plates of the mandibles, they are not separated and the skull is not taken out. As the skull goes back inside the skin, liberties may be taken in cutting the skull, which would not be permissible with a mammal skull.

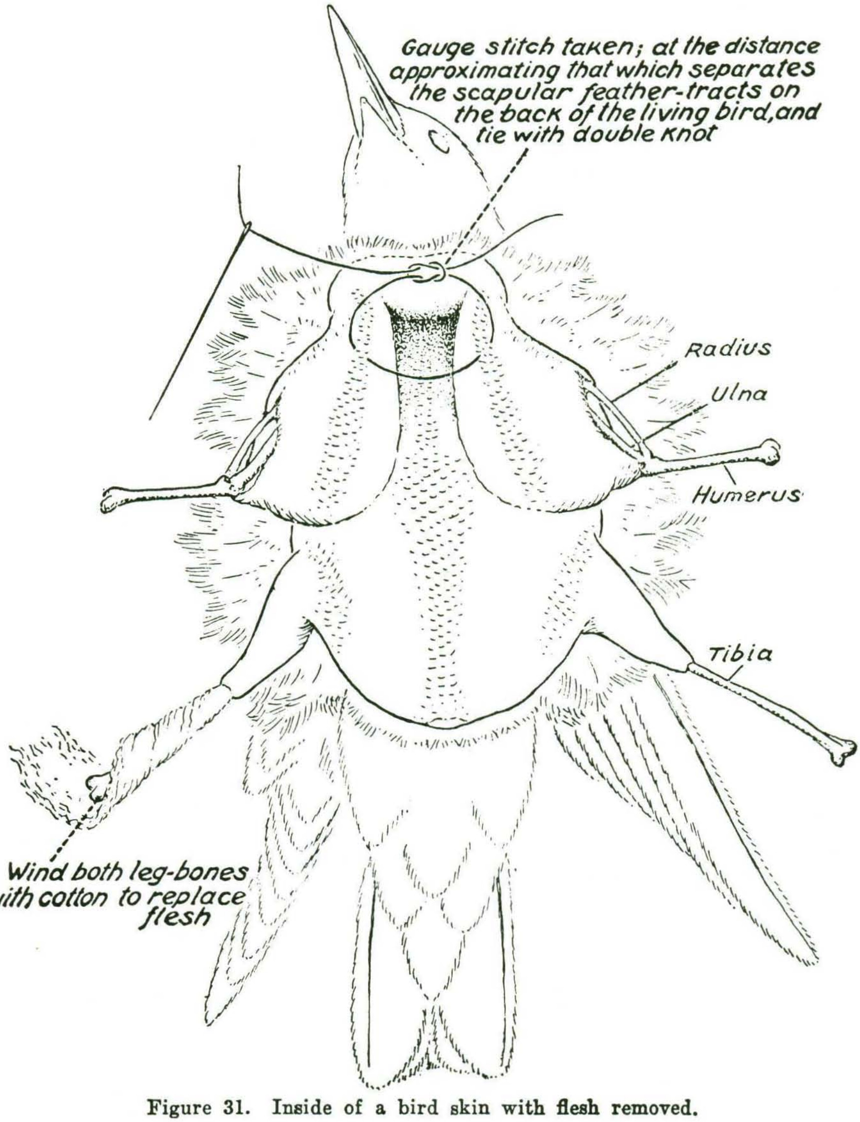

Figure 31. Inside of a bird skin with flesh removed.

With the scissors make a cut directly across the roof of the mouth, below the orbits, but without cutting the lateral supports of either the upper or lower jaws. Then make incisions from each end of the preceding cut within the branches of the mandibles along the under side of the skull as far as the base of the skull. Connect the posterior ends of these cuts by a fourth cut across the back of the skull just above the neck (Figure 30). Pulling the neck from the head will now remove the base of the skull and part of the brain. Large openings should connect the brain cavity with the orbits. The remainder of the brain is scooped out with scalpel or pointed forceps. This method is much better than the old way of hacking off the rear end of the skull and also gives a uniform and dependable orifice for anchoring the neck filling. Cut away any loose flesh from the skull in large birds. In small birds there is little flesh left after brains, tongue, and eyeballs are removed.

Next pull the wing bones inward, peel the skin away as far as the elbow, and clean flesh away from the humerus. Then with handle of scalpel work the skin loose along the upper or anterior side of the forearm (Figure 31). By cutting the tendons at each end the muscles of the

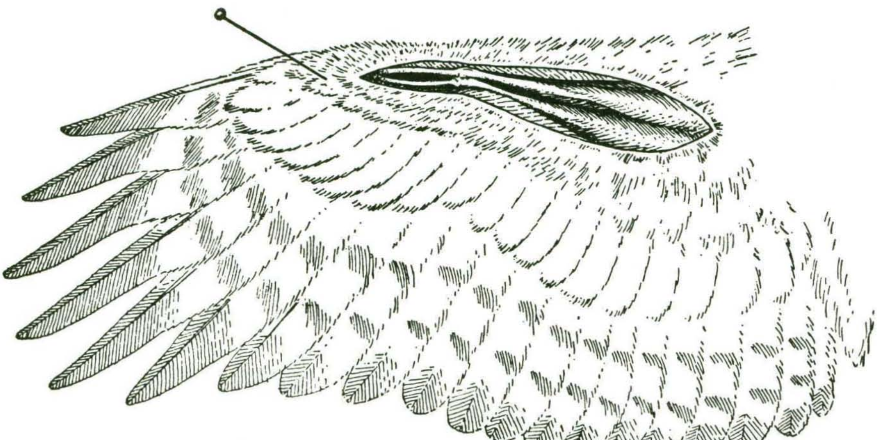

Figure 32. Skinning wing of a large bird.

forearm in all small and medium-sized birds may be removed nearly as far as the wrist (bend of wing). The quills of the secondaries are attached to the posterior edge of the ulna (the lower of the two bones of the forearm) and should never be detached from the bone. Some collectors have a bad habit of stripping the wing down to a point near the wrist, detaching the secondary quills, but this saves very little time and leaves the wing rough, as the quills can not be made to lie smoothly again. In very large birds, the wing must be opened on the outside, on the lower surface, and the flesh cleaned away. It is then poisoned thoroughly to the tip and sewed up with a few stitches (Figure 32).

Turn back the skin from the base of the tail, and cut and scrape away as much flesh and fat as possible. In nearly all birds there is an oil gland just at the base of the tail on the upper side, opening to the outside by a little nipple, from which the bird squeezes out droplets of oil with the beak for dressing the feathers. This oily mass should be scraped away as much as possible, but in very small birds splitting the oil gland and absorbing the grease with absorbent will be sufficient. Do not loosen the tail feathers or cut off the roots so that they fall out. The tip of the tail bone may be left in place between the middle feathers. The skin of the rump is in many cases lined with considerable fat, but is usually thin in this region and the feathers may be loose, so handle this part gently. Any patches of flesh and fat should be removed from the inside of the whole body skin. In small birds the fat is usually found in well-defined areas on the rump, sides of back, breast, and neck. The fatty tissues can generally be loosened at the edges and peeled off towards the centre, and the oily residue soaked up with the cornmeal or sawdust absorbent. Sometimes the absorbent must be rubbed in, scraped off, and renewed several times before the inside of the skin is clean.

Small holes made by shot or punctured in skinning and scraping need not cause any great concern if they are under a feathered area, and a larger rip can generally be sewn together without damaging the specimen very much. Fancy embroidery work on a skin takes time, however, and the necessity for it should be avoided when possible.

Tying Up Wings

One of the greatest difficulties that the young collector meets in trying to " make " a perfect bird skin, is to keep the wings in proper place. The body filling has a tendency to bulge at the shoulders, causing the upper wing coverts to stick up. When the skin is wrapped tightly to correct this tendency, there is apt to be too much constriction elsewhere, spoiling the shape for a prize skin. Wings should always be made fast on the inside of the skin, or they will fall away from the body and are easily broken off. Another useful method recommended by Major Allan Brooks for medium-sized birds is to clean the humeri (upper wing bones) and bind them together by wrapping firmly with a wisp of cotton, which holds the wings firmly in place.

The ordinary method of fastening wings is to slip a cord between the ends of each ulna and radius at the elbow joint and knot it firmly. Draw in the elbow joints until the two humeri lie parallel at the natural position from each other and tie the cords together at this distance (Figure 33). This attaches the wings fairly well, but does not entirely prevent undue bulging anterior to the junction.

A better method of holding wings in proper position is by " the Chapin stitch " (Chapin, 1929, 19). A stitch is run through the forward end of each scapular feather-tract and these are drawn together approximately to the distance separating them in the living bird and tied firmly with a "square" knot (Figure 31). This makes the arrangement of the wing and adjoining feathers vastly easier. Major Allan Brooks, whose bird skins are models for any collector, uses the " Chapin stitch " for small birds only.

Many birds have wings of characteristic shape or outline which are diagnostic of the species in flight, and others have peculiar patterns on the under surface of quills and axillars that are not readily examined in an ordinary dried skin without danger of breaking off the wing. It is desirable for a large study collection to have one specimen each of such species made up with one wing cut off, spread to full extent, and pinned out flat to dry separately from the rest of the skin. The remainder of the skin with the other wing may be stuffed and wrapped like an ordinary study skin and makes a good cabinet specimen. Each part of the skin should carry a label with the same catalogue number, and if necessary the severed wing may be relaxed and attached to the body again later.

Poisoning Bird Skins

If the skin is to be treated with arsenical soap, the soap is better applied while the whole skin is wrong side out. Paint the whole surface of the skin, the skull, the wing and leg bones with a thin wash, seeing that

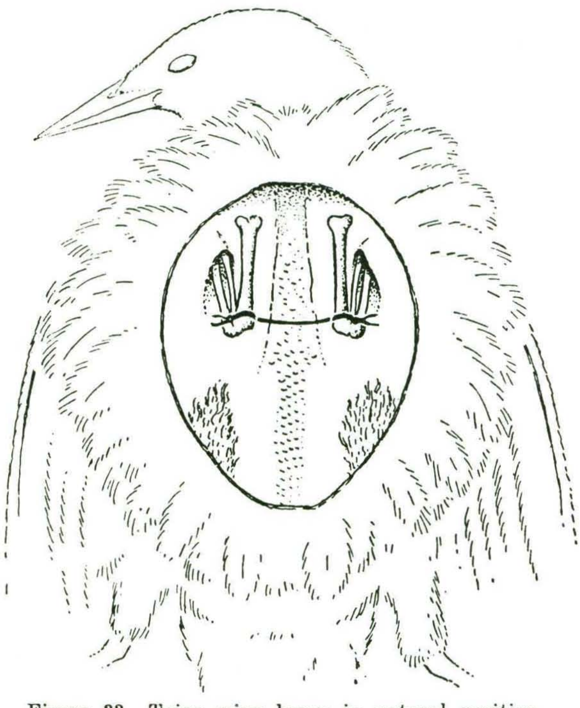

Figure 33. Tying wing bones in natural position.

the inside of the wings and the base of the tail are liberally treated. Then sift fine hardwood sawdust over the whole, damp, poisoned surface to prevent the arsenical soap from rubbing off on the feathers and on the operator's hands. The head skin being soft and lubricated with the soap, it may now be turned right side out by gradually coaxing with the fingers (Figure 34). Holding the skin by the bill and gently pulling at the same time, the head skin will soon slip into place, and by gradually working the skin from the outside the feathers will arrange themselves in their proper places. The appearance of the feathers on crown and cheeks may be improved by thrusting the end of a knitting needle or the head of a large darning needle through the eyelids and working it around under the under side of the skin, stirring up the feather roots.

Cleaning the Plumage

Before filling the skin, look for blood or grease stains on the feathers. If the bird has been cleanly killed and carefully skinned, it should need no further cleaning beyond brushing out dust, cornmeal, and sawdust.

The time-honoured method of cleaning a thoroughly dirty bird skin is to wash the feathers with water containing a little washing soda. Wipe off the excess of water with a cloth, and sponge with turpentine or gasoline so that plaster will not " set " in the damp feathers. Heap plaster of Paris over the skin and rub it into the feathers, beating it out immediately with a flexible switch. The main objection to this method is that the plaster is hard to get entirely out of the feathers, and is almost sure to leave a light powdery bloom, particularly noticeable on dark feathers.

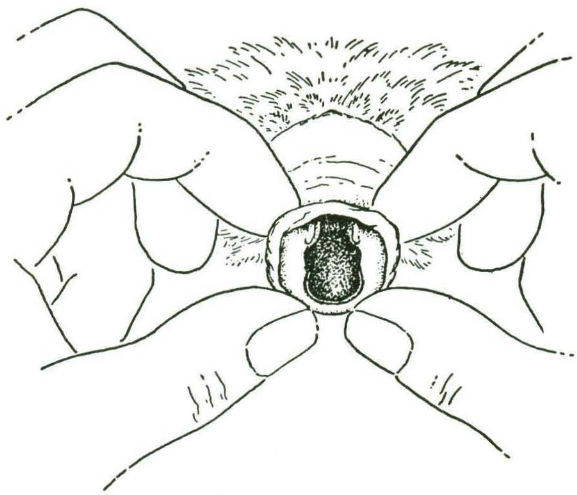

Figure 34. Turning head skin back over skull.

A better method is to clean the feathers with a small, rather stiff brush dipped in clear water, or to sponge them off with a clean wet rag or bunch of cotton, stroking in the same direction as the lay of the feathers. Hydrogen peroxide will remove obstinate blood stains and will not bleach the feathers if dried at once. The feathers must be dried carefully to fluff them out and get rid of the stringy, " drowned rat " appearance. In the field, this is usually done by sifting on cornmeal or other absorbent powder, rubbing it into the bases of the feathers, and lifting the feathers and brushing them lightly with a nail brush. Repeat the operation until the feathers are entirely dry. Fine cornmeal or any starchy meal that does not become sticky when wet is a good absorbent powder. Chapin (1929) recommends a mixture of plaster of Paris and potato starch or potato flour (equal volumes) as the best material for cleaning feathers. Dry, powdered clay or wood ashes may even be employed on dull-coloured skins. The powders may be dried and used over again, any dirty lumps having been thrown away. Shake the powder out of the skin or beat the skin gently, in the sunshine and wind if possible. A pair of small hand bellows is a great help in dusting out skins. Fine hardwood sawdust is very good for cleaning the fur of mammals, and will do very well for absorbing blood while skinning birds, but is unsatisfactory for drying wet feathers, as the sharp, angular points of the wood stick in the feathers, and if the barbules are separated in the process of removing the sawdust, the feathers will never have their pristine smoothness again. In the laboratory, a damp skin may be quickly dried by hanging or holding it close to an electric fan. The use of drying powders will be unnecessary and the feathers will dry smoothly.

Though it is often necessary to use water in cleaning plumage, there are serious objections to using it on white-plumaged birds, as it dissolves fresh blood and the thinned solution stains the feathers. It is almost impossible to clean the winter plumage of a ptarmigan by water methods without leaving some trace of discoloration. Benzine or carbon tetrachloride, or a mixture of both, will clean fresh or dry blood from feathers, and acetone is still better, leaving little if any trace. Acetone of second quality, known as "technical," answers every purpose. Seton (1912) recommends a cream made of benzene and plaster of Paris. Let this dry on the feathers. It dries as a powder and falls off taking the grease with it. Some residue of plaster will remain to be dusted off, but on white plumage it is hardly noticeable.

Hoyes Lloyd (1928) described a method of cleaning large bird skins which had dirt, clay, oil, etc., rubbed into the feathers so that it was impossible to clean them by ordinary methods. After fleshing, the skins were treated with warm water and soap for 10 or 15 minutes in a vacuum cup washing machine, followed by three rinsings in the machine. They were then soaked in gasoline overnight and dried in hot hardwood sawdust. The sawdust was removed by the blower attachment to an ordinary vacuum cleaner.[1]

Windsor (1938) recommends cleaning bird skins by blowing loose dirt away with compressed air; then covering the skin with plaster of Paris and saturating this with carbon tetrachloride, working it into the feathers. When the carbon tetrachloride has evaporated, tap the feathers gently with a round stick until the plaster has been dusted out. Blow the residue of dust particles away as the last operation.

Filling a Bird Skin

After being cleaned and poisoned, the skin should be filled or stuffed to some extent, to show the general relation of the different parts. Skins of small birds should be stuffed to approximately the natural size, but large skins, such as those of swans, geese, large ducks, pelicans, eagles, need be only partly filled and the body flattened dorso-ventrally for convenience and economy in storing. The neck and back of all bird skins should be strengthened with a stiff wire or stick running up into the throat or skull. Thin-skinned birds, such as pigeons, whip-poor-wills, nighthawks, swifts, woodcock, are particularly fragile, and a heavy head renders the neck very apt to be broken off. Skins that are not reinforced with wire or stick are almost certain to have the neck broken off sooner or later as the skin becomes brittle with age.

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[1] *See* page 19.

The writer has seen and tried out several methods of "making skins" in the field, and for many years followed a rather common method with small birds. Put a wad of clean cotton in each orbit either before or after turning back the head skin. Wrap enough cotton around a stiff, slender stick to fill neck and part of body and thrust the end into the back of the skull (Figure 36). Shove a flat bunch of cotton up along the front of the breast into the throat, and put a little bunch into the mouth to fill up under the chin. Wrap the legs and put a little cotton into the abdomen and around the base of the tail, and close the skin with a few stitches. Wrap the skin in cotton or cheesecloth to dry. In the case of smaller birds this method will make a satisfactory skin if properly manipulated, and a good part of the success depends upon proper laying out and wrapping. As a matter of fact, a light and artistic touch will often make a consistently good skin by an inferior or antiquated method. Our hope is to develop a method

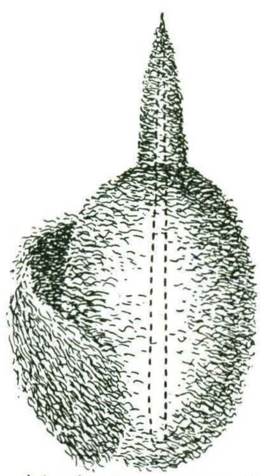

Figure 35. Making artificial body for bird skin.

which if followed carefully will give good average results from the start, and Major Allan Brooks in 1929 demonstrated a method for the writer which is substantially the same as Chapin's (1929) method, with some variations that seem to be improvements.

Brooks' Method of Filling Bird Skins

Wrap cotton lightly around the bones of each leg to form an artificial leg. Roll the neck skin back so that the base of the skull is exposed. On the tip of fine-pointed forceps roll a bunch of fine white cotton into a hard, smooth ball the size of the eye-cavity. Holding it in the forceps, thrust it up through the neck and through the back of the skull into the orbit. A similar ball of cotton is placed in the other orbit, and both are anchored by a pellet of cotton filling the skull cavity and overlapping that in the back of the neck. This neck padding eliminates the skin wrinkle that is frequently formed at the base of the skull when the neck filler is thrust directly into the back of the skull. The neck stuffing is wrapped around a stick a little shorter than the neck and body, or a piece of wire a little longer than the stuffing is to be. The cotton neck should not exceed the natural length of the neck, and the cotton body should be rolled to the approximate size of the natural body (Figure 35). For warblers, spar-

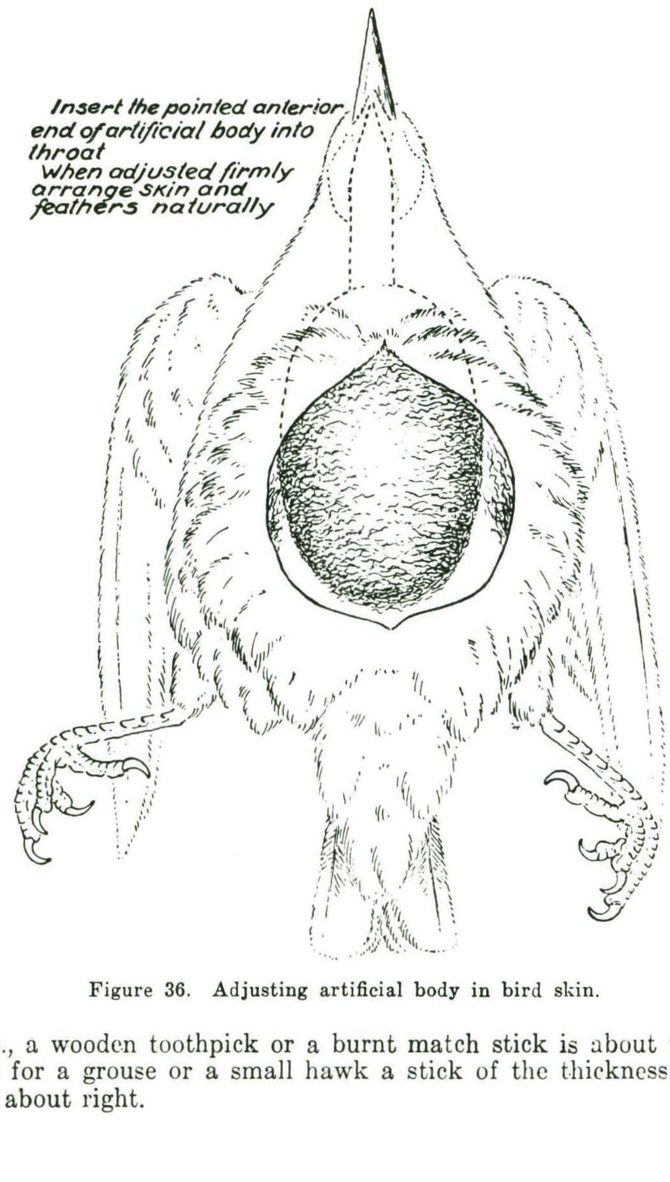

Figure 36. Adjusting artificial body in bird skin.

rows, etc., a wooden toothpick or a burnt match stick is about the proper size, and for a grouse or a small hawk a stick of the thickness of a lead pencil is about right.

Insert the pointed anterior end of the stuffing at the tip of wire or stick, running it up the neck into the throat until the tip appears in the mouth, and pull back the skin of the bird until it encloses the cotton body (Figure 36). Holding the skin with thumb and forefinger on each side of the breast, the plumage may now be arranged properly with the fingers and forceps, the wings folded close to the body and on the back rather than down the chest, with flank feathers overlapping the edge of the wing, and the skin of crown and hind neck pushed forward or backward as required to make the feathers overlap smoothly.

If the skin of the crown or ear-region does not lie smoothly it may be pricked up with a needle point and moved around until the feathers are smooth. When everything is smooth, break or cut off the posterior part of the middle wire or stick so that it comes within the skin. Draw the sides of the opening cut loosely together. In a very small bird the

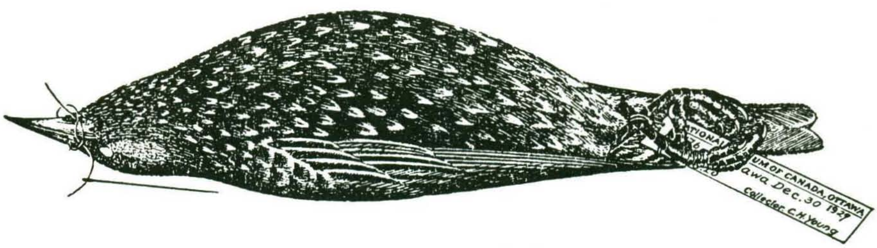

Figure 37. Finished bird skin showing method of tying mandibles and of attaching label.

opening cut need not be sewn up as the skin will dry in place, and the feathers will hide the slit. Most birds should have the opening closed by a few stitches, made from inside to outside. If the edges of the skin have dried too much, moisten them a little inside before sewing.

Bills and Feet

Tie the mandibles together so that the bill is closed in a natural position. A thread may be passed through the nostrils with a fine needle and tied around the lower mandible (Figure 37). If the nostrils are soft, as in nighthawks and cuckoos, the needle is better passed through some other soft part of the base of bill. Care should be taken not to pinch in the sides of ducks' bills; they may be closed without excess pressure if the thread passes over a small pad of cotton placed beneath the lower mandible. The thread may slip off the end of a very thick bill, and to prevent this a fine pin may be thrust between the rami of the lower mandible into the upper mandible. A moistened thread is then used to encircle the beak, posterior to the pin on the lower mandibular side, and with a half hitch on the upper mandibular side somewhat distal or forward of the pin point. The ends of the thread are then brought back to the ventral side, but well forward of the pin for knotting. The pressure by this method is such that it closes the mandibles all along the line and brings the lower mandible forward into its proper position in relation to the upper mandible. Mr. L. L. Snyder, who suggests this modification of the ordinary method of closing thick beaks, states that this method is particularly valuable when the lower jaws are disarticulated from the skull in the process of cleaning it.

Cross the legs about the middle of the tarsi, and attach them by tying on the label firmly with double or square knot, never with a half-hitch or slipknot that may loosen accidentally. The label should be tied with a string merely long enough that the label may easily be turned over. A label with too long a string is a nuisance, as it dangles and catches in its neighbours. Never fail to determine the sex accurately by dissection and mark it on the label before the body is thrown away (*See* page 117). The toes should be spread slightly and not left cramped together to dry, as it may be necessary to compare their lengths later. By the time a small bird is skinned the feet will generally be partly dried and if straightened out will remain in place without further attention. If not, bend them into shape a little later, before they are fully dry.

Wrapping a Bird Skin

All ordinary birds should be laid out in a fairly uniform manner. If the bird has a crest, like jays, cardinals, etc., the head should be turned to one side, usually with the left side uppermost, as skins are usually laid out in trays with heads to left, so that labels are in a position to be read from left to right (Figure 37).

Unroll some fine, long-fibred cotton batting and peel off a thin layer large enough to wrap the bird (Figure 38). Lay the skin in the palm of the half-opened hand and gently press the wings in place, rolling the skin around and arranging wing coverts, scapulars, legs, and tail. If the skin is properly filled and connected, it will retain its shape sufficiently to be moulded symmetrically. Much of the success in "making" a bird skin depends upon skilful handling and wrapping at this stage. Lay the skin, back down, on the thin sheet of cotton and draw one corner

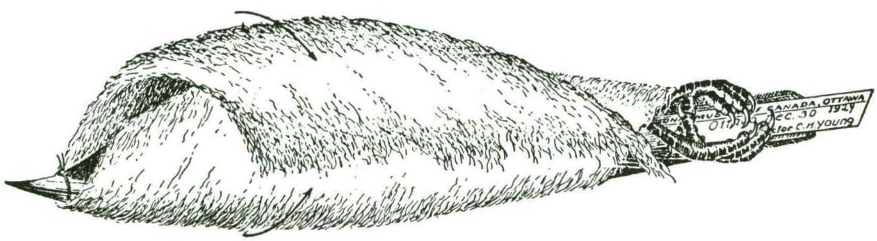

Figure 38. Wrapping a bird skin with cotton.

of the cotton layer up and backwards; and fold the other flap of the cotton to overlap on throat and breast. Turn the skin over and see that the wings come the same distance back and readjust if necessary. Arrange the legs and toes and see that the tail is in line with the body and the feathers spread in a natural position. If the tail has a tendency to become cramped, the feathers may be spread by slightly overlapping each quill on the wrong side of its neighbour until dry. If this is not sufficient, strips of thin cardboard may be placed on the upper and lower sides of the tail and pinned together after the feathers have been spread as desired. If for any reason it is desired to spread the tail fully, run a stitch through the outer edge of each side of the root of the tail and draw them together before closing the skin. In wrapping the skin, slight pressure may be brought to bear at any spot to reduce bulges, and made permanent by merely overlapping the cotton layers. The cotton wrapping is finally drawn together posteriorly and the skin put in a cool, airy place to dry.

Large skins, above the size of that of a pigeon hawk or small grouse, are better wrapped in square sheets of cheesecloth. The cheesecloth is fairly elastic and may be drawn together in the same manner as the cotton wrapping, and held in place by pins where needed. It is easy to see through the cheesecloth and note how the skin is being shaped, and the porous cloth is admirable for letting in air. The cheesecloth wrapping may very well be left on the skin until it arrives at the collector's home port.

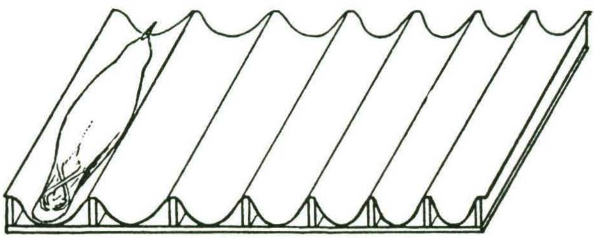

Figure 39. Corrugated drying-board for bird skins.

All bird skins should be wrapped or laid out for drying. The weight of a fresh unwrapped skin will cause it to dry with a flattened back. The old method of rolling a skin in a cylinder of paper is bad, as the operator loses control of the shape, air does not enter well, and the skin dries in a cylindrical shape. Drying in paper cornucopia gives the skin a conical shape anteriorly, and unduly spreads it posteriorly. If the skin is well and firmly made up, and symmetrically shaped, the old-fashioned drying-board, with depressions of varying width and depth, can be made to turn out a fairly good skin if the skin is laid out properly. The drying-board is made by nailing cleats on a board and bending heavy cardboard over these in curves and tacking down (Figure 39). A drying-board can always be extemporized by bending a sheet of cardboard or tin and propping it in place between two blocks or boxes of the proper size.

For stiffening the skins of small birds, Major Brooks keeps on hand a supply of brush pipe-cleaners (cotton-wrapped wire), wrapping cotton around one to form the neck. The tip of the wrapped wire is thrust up into the throat and well into the beak. A thick piece of cotton is laid inside the back and a wisp of cotton thrust into the throat. The centre wire is left a little longer than the neck and body and the posterior end bent into a loop which keeps the body filling from slipping out. Sew up the breast opening, and arrange the feathers, which will fall into place naturally. Major Brooks showed that a properly made skin, with wings fastened, scarcely needs wrapping, and tossed a fresh, unwrapped skin into the air without having it lose shape and symmetry. For this reason he has gone back to the use of the corrugated drying-board, leaving the skin in this for a few hours or a day to round the back. If necessary the skin is then wrapped very lightly with a little cotton, as he considers that tight wrapping causes the bird to lose its fluffy appearance.

The Snyder Form for Drying Bird Skins. Mr. L. L. Snyder, Curator of Birds in the Royal Ontario Museum of Zoology, Toronto, has devised a drying form (Snyder, 1935) that combines in a practical way the good features of the corrugated drying-board with the advantages of wrapping the specimen in cotton. Some of the best features are that the skins are not apt to be mussed or disarranged while drying in camp, and the skins may be left in the forms and safely packed if it is necessary to move camp before the specimens are dry. Another apparently minor feature that is important when large collections are being handled is the slight flattening of the back, which hinders rolling and sliding of skins in storage trays. A beginner can probably acquire the technique of shaping skins more rapidly by this method, although it is well to learn the ordinary methods for use when the "forms" are not available. In making the forms, a graded series of cards, roughly rectangular in shape, with rounded lateral wings, were cut from fairly heavy American cardboard. These were in ten sizes, for specimens from the size of a wren to that of a crow, but all sizes were proportioned the same. Each card was then relaxed somewhat in steam and the rectangular central part was immediately pinned or otherwise held flat on a board that had been equipped with two parallel strips of wood suitably spaced to hold the rounded lateral wings of the card vertical. The spacing of these lateral strips was readjusted after the making of a number of forms of each size. In a few hours the cards were dried out and the lateral wings remained in their vertical position. An imaginary section through the centre of each form presents a rather flat-bottomed U-shape. All of the forms were shellacked and each size given a serial number on the back. A piece of baize cloth was then sized to the inner surface and trimmed. The forms were then ready for use. In the field when a bird skin is all but complete it is laid with the ventral surface downward and the back feathers finally arranged. A thin sheet of cotton of approximately the size of the specimen is then carefully laid over the back. A form of suitable size is selected and slipped over the skin and the whole is then lifted and the ventral surface turned upward. The final adjusting of this surface can be done without the form interfering in any way. The friction formed by the baize and the sheet of cotton tends to prevent the specimen from slipping out of the form when being handled. In addition, a few turns of cops thread around the skin and the form can be made to secure the specimen if such is necessary as in the case of moving camp. A set of these forms telescopes together and a set of six or eight complete or partial sets will ordinarily be sufficient for a collecting trip as forms can be used repeatedly as each successive specimen becomes dry.

Skinning Birds with Large Heads

Some birds, such as many woodpeckers and ducks, have abnormally large heads, and necks so small that the neck skin cannot be stretched enough to slip over the head. In such cases, the neck is cut off and the skin turned right side out. A slit is cut from the middle of the crown along the back of the head and neck, far enough to allow the skull to be skinned and the brain, eyes, and tongue removed in the same manner as with other birds (Figure 40). The necessary stuffing is put into the orbits and the back of the skull and the slit neatly sewn up again. The cut will usually

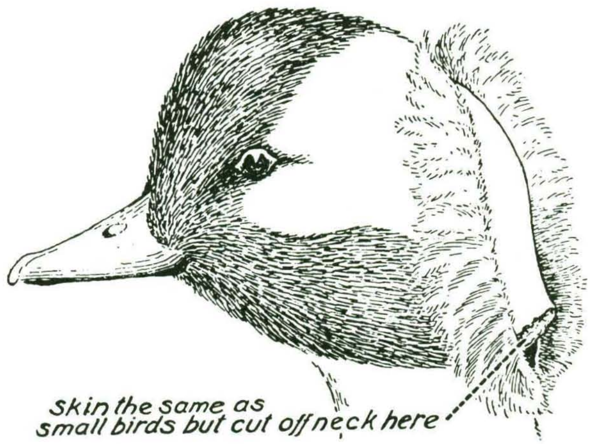

Figure 40. Skinning large-headed birds.

be hidden by the feathers. Some birds that have a horny crest may need to have the head skinned through a slit on the lower side of the throat. Ducks for mounting as "dead-game" pieces, or when the skin is damaged on one side, may have the head skinned by a slit along the defective side.

Wings and Feet of Large Birds

The wings of large birds contain considerable fleshy and ligamentary matter, as well as skin that cannot be reached and poisoned from the inside. A slit must be made along the under side of the wing, from the elbow to a point near the tip of wing where the feathers are fewest, all flesh and tendons must be removed, and the skin poisoned (Figure 32). In the larger birds the skin must be worked loose around the wrist joint (bend of the wing) so that the poison can get in. The slit may be closed by a few stitches, but not tightly enough to keep the air out. The upper arm bones of large birds should be wrapped with cotton to keep them from contact with the skin while drying.

Major Allan Brooks does not consider it necessary to either tie or stitch the wings of large birds in place, but just fills out around the ends of the upper wing bones to the natural size and lays them in position without tying. This filling enables a good display of the scapular feathers to be made. After a large skin is thoroughly dried, the tension of the thick, strong skin is sufficient to keep the wings in place.

The feet of large birds also dry slowly, and the skin may decompose, loosening the scutes or scales that cover the tarsi and toes. Therefore, the tendons and any small muscles in the tarsus must be taken out. The tendons of the tarsi have been loosened at the upper end in skinning the leg. The lower ends of the tendons come together in the sole of the foot, and by cutting a longitudinal slit in the median pad of the sole and pushing the tip of heavy forceps or pliers underneath them from the side, the tendons may be pulled out by main strength. The ends attached to the toes may be grasped with the forceps, pulled out, and cut off as far inwards as possible. If necessary the under side of the toes are also slit open.

Herons and other large wading birds should have another longitudinal slit made, on the *inner* side of heel joint and the skin separated as far around the joint as possible, and poisoned. With a cotton swab on the end of a wire or long forceps, force as much preservative (either wet or dry) as possible inside the skin where the tendons have been pulled out. Mr. Charles H. M. Barrett suggests that filling up the space left after the tendons have been pulled out makes a very much better appearing leg than the shrunken and dried-up leg usually seen on large bird skins. He replaces the tendons with split pieces of bamboo, which is springy and tough and can be split very thin, rounded at edges, and easily pushed up the leg sheath. Bamboo splints may also be used to advantage in mounting birds.

The legs and feet of young gulls and hawks are soft and pulpy, with much watery serum, and need to be opened to dry out. They should be poisoned well or they may become offensive before they become dry and may lose some of their scales. The feet of pelicans and large vultures are difficult to preserve in a warm climate, and it is recommended to slit the skin down the whole length of the tarsus and the underside of the toes to the last joint, to remove the tendons, and to poison thoroughly. The growing quills of young ducks, geese, hawks, owls, and other large birds as they are emerging from the pin-feather stage are in many cases filled with water and blood, so that they are slow in drying and are apt to become fly-blown. These soft young quills should be carefully slit on lower side near bases, the liquid soaked up with blotting paper or other absorbent, and preservative applied to the quills. It is also well to paint the bill, feet, and legs of large birds on the outside with arsenic water, sodium arsenite, or DDT for protection against insect pests.

In mounting either adult or young birds that have large, soft, pulpy feet, trouble is caused by shrinkage and distortion in drying, and a more perfect reproduction for exhibition may be produced by mounting the fresh bird at once and then making plaster casts of the feet in place before drying. The natural feet are then replaced by coloured wax models. Where material is not available for casting feet, combs, or wattles from fresh specimens, the parts to be cast may be cut off and preserved temporarily in a solution of 20 per cent glycerine in water with a little carbolic acid.

Making Skins of Large Birds

As series of large birds are bulky and take up too much room in shipping and storage, the skins are reduced in size as much as is consistent with exhibiting all parts of the plumage. The body is preferably made of excelsior or coarse tow wound with heavy thread or twine, and made smaller than the real body; and the body is flattened a bit after the filling is put in, so that the specimens will not have to be stored in an abnormally deep tray or drawer. A made-up skin is difficult to ship or store if more than about 2½ feet in length, and large birds with long neck, legs, or feet will need to have these shortened by bending; a wire instead of a stick must be used in the neck. Large herons may have the neck bent into a graceful curve (Figure 41), but small heron's necks may be stiffened with a stick and treated in the same way as ducks. If it is necessary to bend the bird's neck back, run the neck along one side of the body outside of the wing, and not down the middle of the back or abdomen. Large and irregular skins cannot be wrapped wholly in cotton, and strips of paper or loose-meshed cloth pinned around them will help to retain the shape of the skin until dry.

In hawks, the sides of the crop should be brought together with a stitch from the inside. This prevents the usually untidy appearance of throat and upper breast in hawk skins.

Trouble is often caused by twisting of the breast feathers, particularly in water birds with short contour feathers, causing a groove to appear along the median line of the breast. In cleaning the skin it is important to break up the fibrous tissue around most of the feather roots with a pointed bone scraper or similar tool, for about one-half inch back from the belly cut. On freshly skinned specimens, according to Mr. Charles H. M. Barrett, the feather twisting can be prevented in most cases by moistening and relaxing the skin with water before sewing up the cut. It is also better to make the stitches through the skin near the edge but not running over the edge. This draws the fleshy edges of the skin together and prevents the stiff contour feathers from being cramped inwards as the skin dries.

Brooks' Method of Making Duck and Goose Skins

One of the common older methods of reducing the length of duck and goose skins was to bend the neck and lay the head along the back or breast. This was an unsatisfactory way, as part of the plumage was hidden and the neck was frequently broken by lifting it up to look underneath. If the neck has to be bent back, it is better to lay the neck along one side of the body. The duck and goose skins prepared by the method of Major Allan Brooks may well be taken as models of neatness, convenience, and compactness (Figure 41). The neck is extended, but shortened by curving. A strong, stiff stick is used for the foundation of the neck filling and a small ball of cotton is made fast around the anterior end of the stick and the rest loosely wound. The round ball is thrust into the cavity of the skull and held fast by expansion of the cotton. The neck skin is shoved up to make it shorter and looser, bent into an elongated S-curve, and loose strips of cotton are put in if necessary on each side of the stick. A large

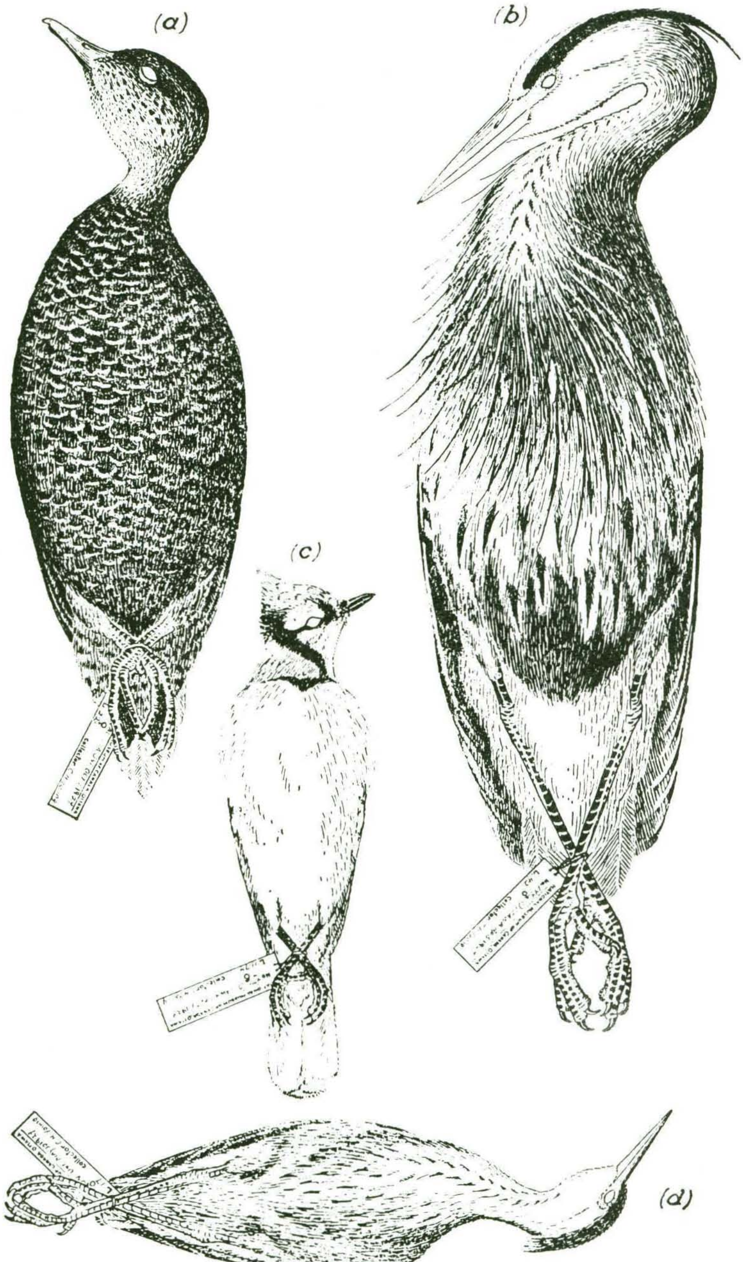

Figure 41. Examples of well-made bird skins: (*a*) black duck, *Anas tristis* (large and thin neck); (*b*) great blue heron, *Ardea herodias* (long neck and legs); (*c*) blue jay, *Cyanocitta cristata* (crested head); (*d*) green heron, *Butorides virescens* (medium neck and legs).

pad of cotton is put under the stick to form the back, and a fair-sized roll of cotton along each side, as described in the method for owls (Figure 42). The legs are wrapped and wings stitched in place as with other birds. The skin of body and neck is considerably flattened dorso-ventrally, and the skin wrapped with cheesecloth to dry. Small herons are made up in the same general manner (Figure 41).

Dill's Method with Waterfowl Skins

Professor Homer R. Dill, Director of Museum of Natural History, University of Iowa, Iowa City, who has had long experience in the field as well as in mounting specimens from old skins, does not approve of bending the necks of duck, goose, or swan skins. He states that bent necks of waterfowl are apt to cause trouble if the skin has to be mounted later, as the bend will show as a crease and the short neck feathers become loose when the taxidermist relaxes a skin that has been dried in bends and tries to work it over. Relaxing bird skins does not work the same as with mammal skins, and some of the feathers may be lost. The waterfowl skins put up in recent years by Professor Dill and some of his pupils are exceptionally fine. He prefers to salt the skin for a few weeks, degrease it thoroughly in the laboratory, and make up the skin with the neck straight in line with the body, with as little stretching as possible, and the head slightly turned to right side and the beak turned a little upwards and forwards.

Treatment of Owl Skins

It is well known that the eyes of owls are abnormally large and forward-looking. The eyeballs are stiffened with bony plates, and project in such a way that it is impossible to regain the natural facial expression if the eyeballs are removed from the skull. After the head is skinned, cut away the transparent cornea in front of pupil and iris, and with small forceps and a wad of cotton force all the liquid contents from the eyeball. The inside may be dried out with sawdust or cornmeal, and preservative should not be omitted. Instead of stuffing out the eyes from the inside, the head skin should be turned back and smooth round lumps of cotton wedged into the front of the emptied eyeballs, through the eyelids. The external ear openings of owls are extremely large and if they have been cut off too close to the surface on the inside may need a stitch or two to close them.

Because the owl looks forward instead of to the side as most birds do, the face of the skin should be arranged to look upward, and Mr. H. M. Laing has kindly demonstrated the superiority of his method. A square cut is made in the *base* of the skull, somewhat larger than with ordinary birds. The supporting stick of the false neck is thrust into the lower posterior part of the skull instead of into the throat, and the stick is retained in place by a large button of cotton tied over the end. A bunch of cotton or excelsior is laid under the stick along the median line and a larger roll on each side. The bunches of filling are held in place by thread wrapped around them, crossing the body (Figure 42). The elbow joints are tied together at the proper distance, or the "Chapin stitch" taken through the scapular tracts. The owl is an awkward, angular bird, with very small body, strong, heavy legs, and long wings, but by making the artificial body carefully and tying the wings in their proper places, the feathers will usually fall naturally into place. This is much to be desired, as the plumage of owls is loose and fluffy and tight wrapping compresses the plumage too much. If the skin is properly made, it will dry very well

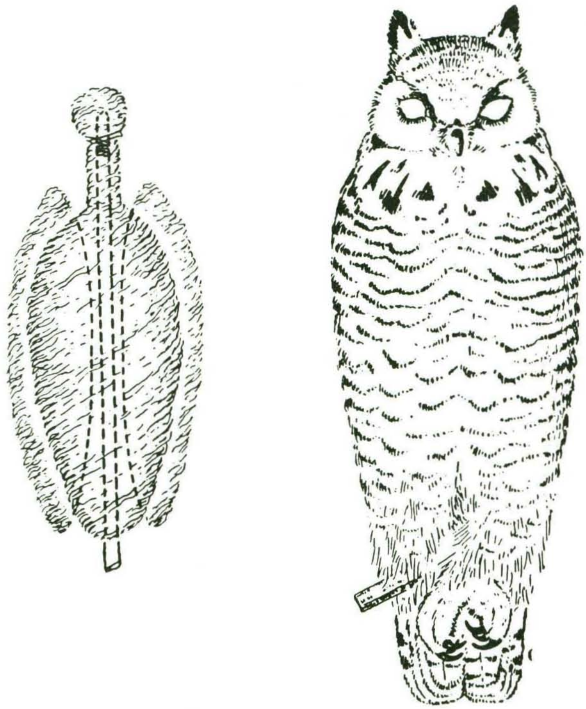

Figure 42. Making up an owl skin.

on a corrugated drying-board (Figure 39), which prevents the weight of the skin from flattening the back of the bird. Wrapping, if necessary, should be put on very lightly, and the skin supported while drying by pads of cotton laid under the wings on each side.

Treatment of Fat Birds

Fat and grease should be removed from the inside of every skin. A skin that is not thoroughly freed of grease may look all right for a time, but within a few months, or sooner if kept in a warm room, the oil will work out through shot holes or rents in the skin, or ooze into the feathers by capillary attraction, and stain and spoil the specimen. Old duck skins in some cases have the breast or back feathers caked with a solid mass of grease-soaked feathers, frequently thickened with dust. This is not the worst result; grease-soaked skins will rot or "grease-burn" by a process of slow combustion, and if ever relaxed for mounting will fall into pieces like wet brown paper.

In ordinary small birds, the fat is usually not excessive, and is found in patches, which may be mostly removed by careful scraping with the scalpel. The residue of oil remaining on the skin is quickly absorbed by sifting on fine cornmeal or sawdust. If the skin appears oily after shaking and scraping off the sawdust, give it another application of the same.

Fat ducks and other water birds often have the whole inner surface of the skin covered with rolls of fat protected by a membranous film. With large, fat birds, the best method of attacking this problem is by shearing off the film and surface fat with a large pair of shears, using care not to cut off the roots of any feathers. This is a tough job, requiring considerable muscular effort, but by practice a sort of sheep-shearing technique may be developed. The fat removal may be begun with the knife, scraping in general away from the tail and in the direction of the head. It is useless to try to scrape in a direction contrary to the lay of the feathers. The roots of many of the feathers extend through the skin into the fatty layer, and the scraping in such cases cannot be very deep or vigorous, as the knife or scraper catches the feather roots and pulls the feathers through on the inside of the skin. The comparatively open spaces between the roots of the patches of contour feathers can generally be fairly well cleaned with the scraper, but frequently the only way that fat can be removed from the feather-patches is by making long slits through the fascia down to the skin between the rows of feather roots. The roots are arranged in more or less regular order and the gashes may be made criss-cross, leaving each feather root standing in a tiny square. During the operations, keep putting on handfuls of dry absorbent and keep scraping it off as it becomes saturated with oil. A piece of hacksaw blade is useful in reaching the depressions between the roots of the feathers that protrude inside the skin. The fascia integument having been opened up by shears, knife, and scraper, and the bulk of the loose fat removed, the time has come to use dry absorbent in mass. Cornmeal or fine sawdust is warmed in a pan, not hot enough to scorch the feathers or skin, and two or three handfuls thrown inside the skin and vigorously rubbed and pounded into the feather roots. The warm absorbent takes up the oil, is shaken and scraped out, and replenished by fresh, warm absorbent until no more oil can be raised and the skin may be considered fairly free of grease.

When nothing better has been available, the writer has cleaned fat duck skins with clean, warm beach sand, and although sand is not a good absorbent it will soften the grease and help rub it out of the skin. Be careful not to have the sand too hot as it will shrink the skin badly, if not burn it. The best cleaned skins that the writer ever sent out were some very fat brant skins which were given to an old Eskimo woman who thoroughly chewed over the whole inside of the skin and swallowed the oil. The skins were left as clean and soft as chamois skin, but it is not always easy to get assistants who like raw goose grease.

If a fat, white-breasted bird has to be mounted, the opening cut may be made along the centre of the back. This keeps the abdomen from becoming stained by grease either absorbed by the feathers in the skinning process, or leaking out ultimately to soil the plumage. The experience of museum curators and owners of large collections is that fat water bird skins retain so much grease after any amount of hard scraping that the grease will come out after a few years. This means that such specimens are good for only a few years, and many museums have had to go to the trouble and expense of degreasing and remaking much of their old material. Some of the old ideas about collecting methods have had to be radically revised, and it is now recognized that in order to preserve fat skins permanently they must be put through a thorough degreasing process with some fat-solvent solution (*See* page 114). Such methods are inconvenient to apply in the field, although if gasoline, benzene, naphtha, or a solution of gasoline-alcohol-turpentine is available, the skin after careful scraping may be dipped and washed in any of these fluids, squeezed out, and dried by shaking cornmeal, sawdust, or other absorbent powder over the feathers and beating and brushing until dry. If the skin has any blood stains, these should be sponged off before degreasing.

Mr. Wharton Huber, of the Academy of Natural Sciences of Philadelphia, who has experimented in this line for several years, wrote the author (November 27, 1931):

> "I recommend salting all fat water birds in the field and bringing them home to the laboratory to be degreased and made up once and for all, for I find even with the great amount of hard scraping that after a few years the grease will come out, therefore, it might just as well be done once and for all where we have proper facilities. . . . . .In the laboratory it is not necessary to salt fresh skins. However, a little salt on the skins and allowing them to rest a few days makes the fat much easier to remove, but this is a matter of option. We are now degreasing all water bird skins whether they are fat or not by this method and are not taking a chance on the grease coming out later on to burn the skin and discolour the label."

Beck's Method for Sea Birds

Mr. R. H. Beck, who has the reputation of being the most experienced and expeditious collector of sea birds now living, makes the opening cut for these birds under one wing, leaving no chance for grease or oil to soil exposed feathers. The fatty masses are rapidly removed by shearing with heavy shears, and the oil absorbed. The filling is made of excelsior or tow wrapped around a stick and inserted through opening under the wing. The wings are held in place by using a long needle to pass a thread crosswise through the body over a few wing quills and back again to be tied over a few wing quills on the first side. The skin is wrapped as usual for drying.

Temporary Preservation of Water Bird Skins

Fat ducks, geese, swans, and sea birds are often obtained in quantity on sea voyages, visits to large rookeries, or under conditions that make it impossible to preserve more than a very few specimens by the ordinary methods. Often there is not time to clean the skins properly and no place to dry the skins if made up, so they are apt to become mouldy, and distorted in shape. To save valuable time in such emergencies, the birds may be skinned out completely and blood stains washed out of the feathers, but without attempting to remove the fat from the skin. In many of the water birds the skin is so thoroughly impregnated with fat and oil that thorough and permanent cleaning is not possible by purely mechanical means, and salting in the field and subsequent degreasing in the laboratory or shop are now considered the only satisfactory means of handling them. Though the salting process is most suited for water birds, it may also be used for other large birds.

Rub fairly fine table salt into the whole inside of the fresh skin, including head, legs, and inside of the wings, forcing as much salt as possible into the skull and around the base of the bill, and fold the skin lengthwise with the flesh sides in contact. If the skin is large and rather damp, it is well to salt and leave it open to dry a bit before folding up. The salted skins should be kept by themselves, but may be packed like sardines and in cool climates will come through in good shape and may be cleaned and made up into good skins even several months later. It is advisable, in order to get the best results, to degrease and make up the salted skins as soon as practicable. This method has been used with success for several years by collectors for the National Museum of Canada, with some improvements in relaxing and making up skins suggested by Huber's experiments (1930).

Relaxing and Making up Salted Skins

The salted skins are immersed in a bucket of water for a few hours to relax them thoroughly, then held under a faucet of running water to wash out all the salt from skin and feathers. It is important to wash out all the salt or the feathers will remain dull or encrusted. When the skin is soft enough to turn inside out, scrape off all the fat with knife or scraper, as described in preceding pages. When the skin is apparently free of fat immerse it in a solution made in the following proportions:

Gasoline, 2 gallons.
Alcohol, 1 pint.
Spirits of turpentine, 4 ounces.

Huber adds in letter (1931):

"The skins either when they come off the bird or mammal or come out of the soaking in water are practically impervious to gasoline, therefore, by using a small amount of alcohol, this mixes with the water in the skin and allows the gasoline to penetrate and removes the grease. The turpentine also assists the gasoline in removing grease, but the main use of the turpentine is to restore to the hair or feathers the iridescence often found in them, for I found, for instance, in wood ducks degreased without the turpentine that the feathers had rather a dead look, while those degreased with this small amount of turpentine had their full iridescent effect."

Immerse in this solution for 24 hours, then squeeze out carefully with the hands until as much of the grease as possible is removed. The first soaking will bring most of the grease to the surface and a second immersion and thorough rinsing in a fresh solution is necessary. These solutions can be used until they become laden with grease when they should be thrown away and a fresh solution employed.

Huber's (1930) method of drying the feathers is by shaking the skins in a tight box or revolving drum with a quantity of hardwood sawdust, preferably maple sawdust. Never use oak sawdust as it will stain the feathers. Take out the sawdust and put in fresh sawdust until the feathers are comparatively dry and fluffy, after which the sawdust is shaken and blown out. Our museum taxidermists have found difficulty in removing sawdust without disturbing the sheen of the feathers, and find it better to dry the skins in front of a strong electric fan. Huber suggests that a vacuum cleaner may be used for a blower by hooking the nozzle on the reverse end and putting a cork with a small hole in it, in the end of the nozzle, in order to reduce the volume of air current and make it stronger. If the skin becomes too dry for making up after going through this process, it may be moistened by leaving it in a damp box for an hour or so. After the skin has been poisoned, it may be made up in the usual manner.

Mr. George E. Hudson, Department of Zoology and Anatomy, University of Nebraska, Lincoln (1935, page 103), describes a method of degreasing bird skins that he considers preferable to the method described by Huber. After scraping the fat carefully from the skin, the skin is made up as usual and allowed to dry in its cotton shroud for about 6 weeks. Several skins may then be degreased simultaneously in a vat or wash-boiler containing several gallons of white gasoline, where they are allowed to remain for several weeks, removed and drained several times, and placed in fresh gasoline. On final removal it is drained head downward for about an hour, after which the skin or skins are placed on a flat pile of newspapers, which are allowed to soak up the gasoline overnight. The newspapers are changed until the specimens are thoroughly dry, whereupon the process is completed. The feathers may require a little stroking to restore them to their former fluffiness, but this requires only a few minutes in the case of well-made skins. Mr. Hudson believes this method far superior to degreasing freshly skinned birds in gasoline, as the plumage is not so much disturbed, and furthermore, gasoline will not penetrate a fresh wet skin as well as it will a dry one, even though alcohol is added to increase miscibility in the former case. He also recommends that turpentine be used in much smaller proportions, if at all, in the degreasing formula given by Huber, as too much turpentine may give the feathers an unnatural gloss.

Degreasing Old Skins

Old, dirty, and grease-soaked skins of either birds or mammals may be cleaned and freed from grease if they are not so decomposed or "burned" by action of the fatty acids that they come apart in the operation. The general method of treatment is the same as for freshly salted skins. Soak the skins in water until partly relaxed, remove the stuffing, and scrape away any large pieces of fat. Immerse in the gasoline bath for at least 24 hours. Very dirty and greasy skins may need two or three days soaking in gasoline, and they should be squeezed out several times a day and the bath changed until it is comparatively clean after squeezing. Mr. Huber recently sent the writer a manuscript addition to his 1930 paper:

> "To degrease a small mammal skin without removing the stuffing: Immerse in clean gasoline for twenty-four hours, draining out several times by holding the tail down and gently shaking; this will remove greasy gasoline from inside. Rinse carefully in clean gasoline. Bury the skin in hard maple sawdust, occasionally brushing the adhering sawdust off with a soft toothbrush. Then bury again until dry. Small bird skins may also be cleaned in this manner.

Hudson (1935) states that made-up mammal skins may be degreased more readily with gasoline than freshly killed specimens, as described in his method given above for degreasing bird skins, adding that the use of gasoline has the added advantage of making the skin less attractive to dermestids.

Hoyes Lloyd (1918) described a method of extracting oil from old bird skins by an apparatus in which ether $(C_2H_5)_2O$ is vaporized and the specimen is repeatedly washed and soaked in freshly distilled ether until not a vestige of fat remains. The ether quickly evaporates and the colour of the feathers remains unchanged. This method was tried out in the National Museum of Canada using a non-explosive solvent, carbon tetrachloride, and the experiment was successful.

The writer has successfully cleaned greasy "made-up" skins of bats and flat skins of other small mammals by suspending them in a glass bottle or jar of carbon tetrachloride for a day or so, and then drying the skins between sheets of white blotting paper or bunches of old newspapers. The hair comes out beautifully clean, but is improved by a little brushing.

Grease floats to the top of the heavy carbon tetrachloride and may be skimmed off, whereas grease usually sinks in the gasoline solution, and the top layers may be carefully decanted off. If the residue is dirty it may be filtered or strained through chamois skin.

Remaking Old Bird Skins

J. Dewey Soper (1943) has perfected a quick and easy method of remaking old bird skins that do not require thorough degreasing. The stitches are severed and as much filling removed with forceps as can be taken without tearing the skin. A small quantity of warm water is injected with a small syringe into the neck and skull through the ventral opening. The body skin in then swabbed inside with wet cotton and a small quantity of this wet material placed inside. If care is used the feathers do not come into contact with water anywhere. The skin is then laid away in a closed tin box where it will become soft and pliable in a few hours; usually over night if it can be worked on the next morning. After relaxing, as a precaution to ensure preservation, the wet cotton filling is removed and the inside of the skin lightly dusted with arsenic and borax. The process from then on is simply the present-day one of making up a bird skin, not neglecting to use a stick the length of neck and body on which to wrap the filling cotton. Before or after sewing up the abdominal cut any grease that may have exuded on the belly feathers of water birds should be wiped off with carbon tetrachloride, and this small area is readily dried with fine sawdust and plaster. Mr. Soper states that by the above procedure following relaxation a couple of dozen small birds can be completely restored in the course of a few hours work.

Skinning Downy Young Birds

Young birds in the "downy" or "natal" plumage are of great interest and the young of many species are rare in collections. Most species of shore birds (Limicolae) and some of the ducks and geese breed only in the far north, in regions that ornithologists seldom visit. Even in more accessible localities, the young birds are seldom seen. They begin to change into the feathers of the first autumn plumage within a few days after hatching, and are only obtainable by a collector who is on the breeding ground for a few days at hatching time. Even when a nest of eggs is found and watched during incubation time, it is hard to catch the young, as they run away and hide as soon as hatched. Messrs. H. W. Brandt and O. J.

Murie collected fine specimens of young shore birds in Alaska by taking the eggs as soon as they became "pipped" and hatching them in a warm place, but this method is not always practicable.

Downy young birds are somewhat difficult to skin as the skin is very tender. The down is also hard to clean if soiled, and it is not easy to remake the skins. Major Allan Brooks advises that before attempting to skin a "downy", the collector should make a careful examination of the pterylosis or arrangement of the feathers in definite areas of growth (*pterylæ*, or feather tracts). If the specimen is overstuffed, the markings will be distorted and the short down feathers will not overlap the bare areas. A downy specimen will usually make up fairly well if not overfilled, especially around the head and upper back, and the skin worked forward and not pulled back.

Make the opening cut along one side of abdomen, and not along the middle, to avoid wetness which ensues if the umbilicus is opened. If the specimens are to be used in a mounted group, it is well to mount the skins in the field, wiring the legs and anchoring them in a small piece of cork. Pad with bits of cotton. The orbits should be filled with cotton, and the colour of the iris carefully noted so that properly coloured glass eyes may be put in later. Also note carefully the colour of bill and feet, as in some species the colours of these parts in juvenile specimens are unknown to ornithology.

The sex of young ducks in the downy stage may generally be determined by the much greater size of larynx in the males. If several specimens are in the brood comparisons may be made and verified if possible by dissection.

Determining Sex of Birds

Determination of the sex of a bird from plumage and other external characters can not be depended upon. Male and female of any species normally have different plumages at certain seasons, but young birds and adults in winter plumage are often hard to tell apart. Female birds may occasionally develop male secondary sexual characters through pathological conditions. All bird specimens should, therefore, have the sex verified by dissection.

After the skinning is completed, and the body removed, cut open the left side from the vent to the anterior border of the ribs. Push the intestines aside and look for the sexual organs, which lie in the small of the back close to the backbone just below or ventral to (on the abdominal side) the anterior end of the kidneys. The kidneys of birds differ somewhat from those of mammals, being large, soft, elongated, dark brown, irregular-shaped masses filling the concavity of the sacrum.

The writer for years followed Hornaday's method (1892) of opening up the body by a slash across the wall of the abdomen, then breaking the small of the back, which snaps off very easily when the front of the body is grasped with one hand and the tail part with the other. The intestines will at once fall forward without any further handling, the rectum having been severed early in the skinning operations. The intestines being out of the way, the testes or ovary should be plainly exposed. The best feature of this back-breaking method is that the sexual organs lie just back of the break, and the young dissector knows just where to look for them. However, as there is sometimes congestion of blood in the lumbar region, breaking the back may cause effusion of dark blood, which obscures the small organs, and it is generally better to follow the more careful method described above.

The male organs (testes) are two in number, normally white or yellowish in colour, but sometimes darker, and lie side by side. They are rounded or ellipsoidal in form, and for some time before, during, and after the breeding season, they become very large and conspicuous and can not be mistaken. In young birds and in winter specimens the testes may be extremely small (less than the size of a pinhead in some species), but can usually be distinguished unless broken up by shot wounds or decomposition. In the small, shrunken state they may not be larger than grains of sawdust, but the latter will be unattached and can be wiped off. Do not confuse the testes or ovary with the adrenal bodies, which are smaller and flatter, yellow or orange in colour, and lying farther forward *in* the anterior border of the kidneys (Figure 43).

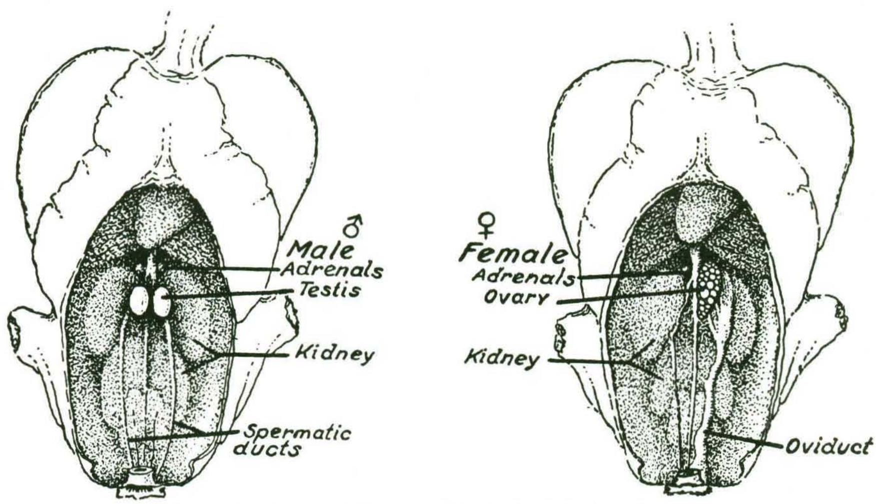

Figure 43. Determination of sex of birds by dissection.

The female organs usually consist of a single ovary, lying a little to the left side on anterior surface of the kidney (Figure 43). A vestige of the right ovary may be present. During the non-breeding season the ovary is a pale-coloured mass of small undeveloped ova, irregular in shape and somewhat flattened. The granular structure, or minute divisions, are usually visible to the naked eye, but often a magnifying glass is necessary to identify the organ. The adrenal bodies are situated as in the male. During the breeding season there is no difficulty, as some of the ova become enlarged to form the yolks of the eggs, the ova being of graduated size according to their maturity. At the same time the oviduct, a tube leading from the neighbourhood of the ovary down the left side to the cloaca (vent, or common outlet for rectum, bladder, and generative organs in both sexes of birds), becomes enlarged, contorted, and whitish in colour.

The best way to become familiar with the anatomical conditions is to carefully cut up a rooster and a hen, because the sex is known in each case, and the organs are large enough to be easily observed. The sexual organs in all other birds are substantially the same except in size.

The comparative size and development of the reproductive organs show the sexual maturity of the bird, as well as the season of reproduction, and should be noted on the label: "testes (or ovary) enlarged," "somewhat enlarged," "slightly enlarged," or "not enlarged." In small species, this is still better indicated by an outline sketch of the true size and shape, or in large species by the measurements in millimetres. Any abnormalities should be noted, as they often have an effect upon the plumage, or other secondary sexual characters.

Finally, never fail to put a question mark after the sex mark on the label if there is the slightest doubt about it.

Determining Age of Birds

Some large birds do not attain their full development until their third or fourth year, and the age may be told from the condition of the plumage. Most birds, however, attain maturity the first spring after hatching, when they put on the first nuptial or adult spring plumage. As many adult birds moult into winter plumage, which is essentially the same

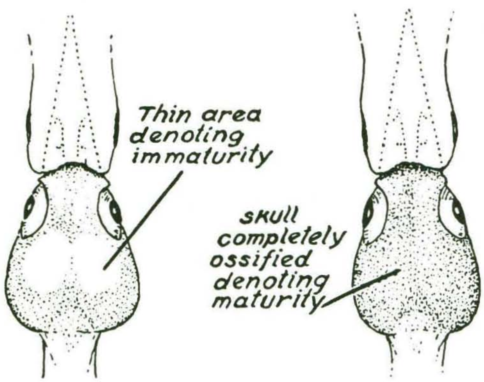

Figure 44. Determining maturity of birds by the skull.

as the first winter plumage of young birds, it is of interest to know the age of such birds when possible, marking on the label "ad." (adult) and "juv." (juvenile) or "im." (immature) for any bird that is in its first year.

In buying poultry it is possible to tell a young bird by the smoothness of the legs, or by feeling the posterior end of the sternum (breast bone), which ossifies late and has a cartilaginous rim in young fowls. The late C. Lehn Schiøler, a Danish ornithologist, claimed to be able to tell the age of eider ducks to a month, up to the age of 4 or 5 years, by the development of the sternum, but very little is known about the detailed bony development of most species of birds, and the application of osteological data is only practical to a limited extent in the field.

However, with practically all passerine birds (perching birds, the ordinary song birds and their relatives) it can be determined from the skull whether the specimen is a bird of the year or not. As the bird's skull goes back into the skin again, it is obvious that the examination of the skull must be done by the collector in the field.

In the nestling the bony roof of the brain case is very thin and transparent, but in the adult bird it is more opaque or nearly white, being formed of two layers of bone separated by air spaces. When an adult skull is held up to the light all parts of the roof of the skull show fine dark specks. As the young bird develops, the transparent area becomes smaller and the dotted structure crowds in on its edges until after 3 months there is only a relatively small clear area, where a single layer of bone persists (Figure 44). This is usually near the middle, back of the eye, but in some families of birds, as in the swallows, it may be farther back. After about 6 months this sign of immaturity is lost. A very few exceptional species of Passeres retain a thin area in the skull throughout life, and woodpeckers and some other families have a different development.

Information about this character of the skull is desired for all species of birds, and the collector should always note it on the label. The writer puts down "skull clear," denoting immaturity, or "skull granulated," denoting adults; others write "s. n. o." (skull not ossified), or "s. o." (skull ossified). The science of ornithology is becoming more exact and technical every year, and the student of bird skins wants his data where it can be readily found, and not buried in some absent collector's notebook. It is not desirable to increase the size of the label, but if there is not room enough for the accessory information demanded, tie on a second label.

Stomach Contents

The economic value of a species largely depends upon its food habits, so the crop and stomach of a skinned bird should always be examined, and preferably preserved. A field investigation can seldom be complete, as the average ornithologist is seldom able to name all the other animals, insects, seeds, or fruits that have been eaten by the bird. If a bird of prey has been eating other birds or mammals, the material in the crop can often be determined at sight, and a rough estimate may perhaps be made of the insect or vegetable contents of the stomachs of other birds. It is advisable to make a routine practice of removing crop and stomach, tying both ends, labelling with the number of the bird specimen, and putting it into a bottle or friction-top can with alcohol or formalin. It is important that a crop or gullet containing food be kept with the associated stomach. A season's collection of small bird stomachs can usually be contained in a quart fruit-jar or can. The contents of stomachs of small mammals are usually much more difficult to determine than those of birds' stomachs, as most mammals have teeth and chew their food before swallowing. Mammal remains can be identified by the presence of teeth, feet, claws, and hair, and birds by feathers, feet, claws, and bills. Considerable information has been given by Dixon (1925) and Hamilton (1930), and much more attention should be given the subject by both ornithologists and mammalogists. (*See also* "Directions for preservation and care of material collected for food habits studies"; U.S. Biol. Surv., Wildlife Research and Management Leaflet BS-29, Dec. 1935.)

In the case of reptiles it is well to save the entire digestive tract. It is often impracticable to save the entire stomach contents of large herbivorous mammals, but samples may be taken from several points in the anterior stomach or rumen and thoroughly mixed before removing the part to be saved. This with some of the larger and more diagnostic bits of food may be placed in a cheesecloth bag or wrapper and then handled in the same manner as a stomach. The essential data are the same as for skins, but hour of day should be given, and if trapped what kind of bait used.

After the stomachs come in from the field, they are sorted by species, and each species is preserved in a separate bottle until the stomachs can be examined in detail. A book or card catalogue should be kept, with the data accompanying each number.

Temporary Preservation of Fresh Specimens

During the average Canadian winter, birds and small mammals may be kept for considerable periods if the body can be frozen solidly before it starts to spoil. If well wrapped in several thicknesses of paper and stored in a cool place out of the sun it will usually not thaw out even if the outside temperature is above freezing during the day, and will remain in fair condition for skinning except that the legs, feet, and wings may dry somewhat. Specimens frozen solid and well wrapped in paper will also stand 3 or 4 days' shipment by mail or express without spoiling. There is considerable variability in the keeping qualities of animals, but birds usually keep better than mammals; rodents and herbivores better than carnivores; and insect-eating species (moles, shrews, and bats) are the worst of all.

In warm weather, if more specimens are obtained than can be skinned immediately, an ordinary household refrigerator will help, but will not keep the temperature low enough to do for more than a day or two. Small specimens that are not malodorous may be wrapped in paper and kept in a home frigidaire for some time. If placed in a cold storage warehouse, specimens will keep for weeks in fair condition. A well-equipped museum should have one of the modern types of electric refrigerators or a refrigerating room where specimens may be kept in a frozen state until the preparators are ready to work on them.

It is best to skin all birds and mammals as quickly as possible and poison them in the usual way, then place a small bunch of damp cotton inside and bring the edges of the skin together. Place the skins in a tight box with a wad of cotton, a slab of plaster, or some wet sand, saturated with water and a little lysol to arrest decomposition. Skins of birds and small mammals will remain soft and pliable for a week or longer, and may be made up at leisure. The feet and legs may dry somewhat and may need to be relaxed by wrapping them in wet cotton. The writer has found a damping-box, or a 2-quart fruit-jar with some wet cotton in the bottom, convenient for partly relaxing dry bat skins so that the fingers may be straightened out and examined. Leaving the specimens in the damp air in a tight receptacle over night is usually sufficient.

Drying Skins of Birds and Small Mammals

Small skins will usually dry very well in a few days in a normal living temperature, and where stove or furnace heat is available they will dry fairly rapidly if placed on shelves or suspended in trays (preferably with wire screen bottoms) where warm air can circulate freely. Specimens should not be exposed to hot sunshine or placed where the heat is so great that it feels uncomfortable to the hand.

Along the seacoast, particularly in the Arctic regions, it is often a problem to dry skins during prolonged periods of damp or foggy weather, especially if the collector is living in a tent. On a vessel the collector may be able to dry his specimens in the fiddley or hang them over the galley stove. The writer has had some success in drying specimens in a tent by using a "Cambridge tin," a tin case with sliding trays and a tight cover fitting into grooves and secured with clamps. The Cambridge tin may be left open and set near the camp stove as a sort of drying oven, but needs constant watching and adjustment to prevent it from becoming overheated. The Cambridge tin, or any sealed tin box enclosed in a wooden case for protection, has some use in shipping equipment that must be kept dry, such as photographic films, chemicals, etc., and is valuable for storing or shipping thoroughly dry specimens, because mice and insect pests are effectively kept out. On the whole, the Cambridge tin is unhandy for a collecting box in the field and the screen-windowed box (Figure 1) is much preferable.

If skins are not thoroughly dried before packing, as is often the case in damp weather, the packing case should be painted inside with carbolic acid or have plenty of naphthaline crystals scattered over the specimens. A coating of greenish or bluish mould may form on feet and bills as well as on feathers or fur of imperfectly dried specimens. If the process of growth has not gone too far the mould may usually be wiped off with a damp cloth and the specimen dried and dusted clean. Sponging with a bit of cloth dipped in alcohol, gasoline, or benzene will aid in killing mould in wrinkles and crevices. Mould only occurs on specimens that have been kept for a time in a damp place, or on infected specimens that have been put into tight cases before being thoroughly dried. In most parts of Canada this does not cause the serious difficulties met with in the more humid parts of the tropics.

Packing Specimens

Under ordinary circumstances it is better to pack specimens in wooden boxes or crates through which there is some circulation of air. Mice and the larger insects may be kept out by lining the packing cases with cheap wire fly screen, or by tacking strips of screen over the cracks. A liberal quantity of naphthaline sprinkled on the specimens and in the bottom of the box will usually keep out the pests, whether of animal life or of vegetable fungi. Corrugated fibre-board cartons are coming into general use, and are more easily trimmed to size needed.

The best way to pack small skins for shipment is in flat pasteboard boxes, such as shirt boxes, in which case the skins need not be wrapped individually, but may be laid closely together on a sheet of soft cotton with wisps of cotton laid in the crevices between specimens, and a layer of cotton laid over the lot before placing the cover on the box. If desired, the pasteboard boxes may be purchased from the manufacturers in knockdown form, but usually the small number needed may be obtained from local dry goods or clothing stores. If the smaller boxes are not available, the skins may be packed in bulk, but each skin should be wrapped in a piece of paper before it is finally packed. A good method is to roll each skin in a cylinder of paper (preferably heavy, sized magazine paper) and tuck in the paper at each end of the roll. A thin layer of cotton should be placed on the bottom of the box, and a layer of cotton between each layer of skins. If the box is large and the skins rather heavy, the layers may be separated by sheets of cardboard, pasteboard, or a folded newspaper, or by a thin board supported by nails driven into the inside of the box. If a large skin has to be packed with smaller ones it should be pinned down or separated from the others by a partition, to avoid crowding. Scatter naphthaline flakes in among the skins as they are packed.

External and Internal Parasites

It is well known that most species of animals are subject to infestation by external and internal parasites, some of which may be peculiar to a single host species, some common to a genus or a family, whereas others may have a life history running through two or three unrelated species. Some species such as muskrats and ruffed grouse, are known to harbour twenty or thirty species of parasites. Many of these are comparatively unimportant to the host, but others are directly injurious to the health, or may carry germs of disease that may be fatal. Increasing interest is being taken in the subject of parasitology in mammals and birds, as parasites have considerable bearing on causes of disease and of fluctuations in numbers of certain species.

As collectors should always be anxious to make their work of the greatest possible practical and scientific use, they are urged to pay more attention to these subjects, which have generally been regarded as unimportant, and to collect parasites from named hosts whenever practicable. Information gleaned from a lot of dead birds or mammals dumped together may not be reliable because external parasites, such as bird lice and ticks, seek fresh fields as soon as possible after the death of the host. Dead mammals in traps are usually comparatively free from external parasites. If the animal is fresh, the parasites may be shaken out on a sheet of paper and placed in small vials of alcohol, or the animal may be placed in a grocer's paper bag with opening tied tightly until time is available for examination for parasites. Birds are usually obtained in a fresh state and may be kept in tightly rolled paper funnels for examination for external parasites. After skinning a mammal or bird, the throat, crop, stomach, and intestines should be examined for internal parasites such as tapeworms and round worms.

Directions for Collecting Animal Parasites

Dr. C. H. D. Clarke, formerly of the National Parks Service, Ottawa, and now chief of section of Wildlife Management, Fish and Wildlife Division, Department of Lands and Forests, Toronto, Ontario, collected during three field seasons for the National Museum of Canada and has kindly prepared the following directions.

Quite satisfactory collections of parasites can be made from specimens taken under most field conditions. The types of specimens are as follows:

(1) *Ectoparasites.* The first requisite is that the host specimens must be separated so that no mixing will take place. Paper funnels accomplish this quite well, but small paper bags are in my experience the most convenient and the most reliable. The ectoparasites can then be picked off at leisure. There is no substitute for actually picking over the fur or feathers. For example, I have always found the small biting lice of *Peromyscus* attached to the host, or in the fur. Human lice have been collected from mummies. The parasites when collected are transferred on the moistened end of a probe, match, forceps, etc., to a vial of 70 per cent alcohol. Some collectors prefer a small brush. Personally, I detach them with the forceps and pick them up by moistening the forceps. In collecting a tick firmly attached to the host it is best to take a piece of skin along with the tick, in order to avoid damage to the mouth-parts.

(2) *Endoparasites.* Occasionally large tapeworms or round worms protrude from the gut, or filaroid worms may be seen in tendons, subcutaneous tissue or peritoneal cavity. Smaller parasites are obtained by washing the contents of the various parts of the gut in water and allowing them to settle, pouring off the supernatant. Two or three washings will remove most of the food material and reveal the parasites. An eye-dropper is convenient for transferring them to vials, if they are too delicate for forceps. Parasitic worms are all satisfactory for study if preserved in 5 per cent formalin.

For the above two types of parasites a stock of small vials with corks, 95 per cent alcohol and full strength formalin for diluting, comprise the equipment. Data for endoparasites should include the locus of the parasite within the host. Labels with full data for all parasites should go in the vials.

(3) *Blood Parasites.* A third type of parasites are those found in the blood. In my own collecting I always make a smear of the blood of each specimen on a glass slide as soon as possible after collecting. This film should be dried in the air and fixed within an hour or two by covering it with absolute alcohol (methyl or ethyl hydrate) for 5 minutes. It may then be stored in a slide box or wrapped in paper, to be stained at leisure.

A supply of glass slides and boxes (1 inch by 3 inch slides) is needed, as is also a supply of absolute alcohol, about 200 cc. for 100 specimens. Blood from a small mammal is best taken from the heart.

(4) Abnormal growths, diseased organs, tapeworm cysts, and organs with parasites in situ may be preserved in 5 per cent formalin in bottles. A supply of small bottles and corks is necessary.

Parasites collected and preserved as described above would yield valuable records and data of much use, particularly if the data accompanying the specimens are complete.

Animal Diseases

If there is an epidemic among any wild species, or an animal appears to be diseased from unknown causes, it is well to make a "blood slide." Put a drop of fresh blood on a clean glass microscope slide, smooth and spread out the drop of blood with the edge of another clean slide. "Fix" it with a drop of pure grain alcohol, and, holding the slide by the edges, dry it by waving in the air. Then wrap the slide in a sheet of clean paper, and submit it to some veterinarian or bacteriologist for examination. The veterinary departments of our colleges or the Health of Animals Branch, Science Service, Department of Agriculture, Ottawa, will usually be glad to get data that will help them in studying animal diseases. (*See also* Shillinger and Rush, 1937.)

Through the courtesy of the Veterinary Director General, Health of Animals Branch, Department of Agriculture, Ottawa, the following additional directions were prepared by Dr. E. A. Watson, formerly Chief Pathologist, Animal Diseases Research Institute, Hull, Quebec (August 18, 1932):

"As regards the collection and preservation of specimens for pathological purposes the following methods of procedure are usually employed:

For gross specimens: immersion in 10 per cent formalin [1 part commercial formalin to 9 parts water], the volume of fluid being several times that of the specimen. [If possible, some of this solution should be injected into the specimen itself.]

For microscopical examination: small pieces of tissue ½ to 1 inch in length and thickness, in several volumes of 10 per cent formalin.

For bacteriological examination: specimen material immersed in 20 per cent glycerine (in sterilized or boiled water) or packed in gauze impregnated in dry boracic acid. Bacteriological specimens should be packed in ice and sawdust and sent in to the laboratory as quickly as possible."

COLLECTING BIRDS' NESTS AND EGGS

The collecting of birds' nests and eggs is a time-honoured schoolboy pastime that has largely fallen into disrepute. Though collections of eggs are not in themselves considered of great value to science, and indiscriminate collecting is generally frowned upon, nests and eggs are essential to museum habitat groups, and are of some interest to the general public. Although a large proportion of the older ornithologists started their careers by birds'-nesting, the study of nesting habits has never received the credit it deserves in bringing to light the various manifestations of life histories, or habits, which reach their culmination in the breeding season. Although many oölogists are not very "scientific," particularly in regard to fine points of identification, and others do not put their valuable observations on record, they have learned that it is necessary to watch and study the *habits* of birds *in life* before the nests may be found and frequently know much more about the real life history of the bird than the skin collector who shoots his specimen at sight. The study of the nest of the bird is also of interest, as the nest shows much more about the intelligence of the bird than the mere egg-shell, a simple physiological product of nature. Much more remains to be done in studying periods of incubation, study of embryo chicks, feeding and growth of young birds, and photographing nesting sites and the parent birds near the nests.

When eggs are collected, full sets or clutches (the full number of eggs laid by the birds) are desirable, and if possible the eggs should be collected when fresh, just at the beginning of the period of incubation. Some species lay at intervals and begin incubating the first egg, so that the young do not hatch at the same time. A few tools are needed: (1) egg drills, from about $\frac{1}{16}$ inch up to $\frac{3}{8}$ inch in diameter; (2) curved metal blowpipe (a straw or section of grass stem may be used in emergency); (3) fine, curve-pointed scissors; (4) fine, curve-pointed forceps; and (5) embryo hook. A small, hard-rubber piston syringe is very handy, but the collector can get along by blowing a mouthful of water through the blowpipe.

A hole is drilled in one side of the egg, no larger than will allow the contents to be emptied conveniently. A hole that is too large is unsightly and weakens the shell. Holding the egg hole down, place the tip of the blowpipe near the hole and blow gently, until the compressed air forces the contents out. If the egg is incubated, the liquid part may be blown out, and by careful work with forceps and scissors the remainder may be pulled out piecemeal. If the egg is considerably incubated, and hairs or feathers are noticed, the job is more difficult, and after blowing out the softer parts, a solution of pancreatin in water may be injected and the egg set in a warm place until the contents are digested enough to be rinsed out. Always rinse out the inside of a blown egg with a mouthful of water forced through the blowpipe or syringed into the shell, before setting it aside to drain on a piece of cotton. Blowing an egg that has a chick in it is a tedious and messy process, and unless the egg belongs to a very rare species, and the collector has the patience and skill to carry through a long operation, the egg is better left in the nest. A well-incubated egg can usually be told by loss of the original pearly tint, dull colour in thin-shelled eggs, and polished surface of shell in other cases. The appearance is rather difficult to describe, but is easily recognized by a person who is familiar with the appearance of the fresh egg. Eggs that have characteristic spots or other markings on their shells should be kept from wetting if possible, as the pigment is often solvent in water when fresh, and the pattern is easily smeared or obliterated.

Oölogists sometimes obtain eggs that are too far advanced in incubation to be blown without splitting the shell. In such cases the embryo may well be preserved in alcohol or formalin and turned over to some embryologist who is interested in the ontogeny and phylogeny of birds.

An egg is totally useless unless the identity of the parents is known. In some cases, the shape or colour of the egg is unique, and the egg can be named by inspection, but in most cases it is necessary to have a good sight identification of the parents, and if the collector is not able or competent to do this, one of the parent birds must be secured and preserved for identity.

To avoid mixing specimens, each egg in the set is marked near the blowhole with soft black pencil with the same number, the A.O.U. number of the bird, and the set mark. The set mark consists of the collector's field number of the set and the number of eggs in the set. A robin would have the A.O.U. No. 761, followed by the collector's field number, and if it was his first set of four eggs, by the fraction $\frac{1}{4}$. The second set of four eggs would be marked $2\frac{}{4}$. If the nest is preserved, the letter N is usually added.

A set of eggs should always be accompanied by an authenticated pedigree in the shape of a data label. The data blank for this should nct be too large, but should have space enough for the A.O.U. number, set mark, number of eggs in set, name of species, locality, date, incubation, identification (parents observed, shot, or named by. . . . .), nest (situation and composition), name of collector, and remarks. The same data should also be kept in the collector's catalogue or record book for future reference, as the label always goes with each set of eggs if they are sold or exchanged.

In shipping eggs, each egg should be separately wrapped with tissue paper or a wisp of thin cotton batting, and each set of small eggs placed in a separate container—a pasteboard box or a tin of proper size, or in a larger box with partitions, and enough loose filling to keep the eggs in place. Large eggs should, if possible, be wrapped and packed in individual compartments, similar to those used in shipping hens' eggs to market. They are usually worth more per dozen than hens' eggs and the collector should be willing to take proper care of them. Each nest should be wrapped separately, with plenty of thread to keep it from falling apart.

Egg collections are usually kept in cabinet drawers, with each set or nest in a shallow pasteboard tray. Loose cotton is rather springy and the eggs may bounce out when the drawers are pulled out. So it is best to line the trays with a section of layer cotton cut to proper size. Many collectors prefer to use medium fine sawdust in preference to cotton wool as bedding in trays, but freshly blown eggs should not be placed on sawdust as the grains of sawdust are apt to stick and the eggshell may break when sawdust is being removed. The eggs may be kept from rolling by bits of cotton at the sides. In museum collections sets of eggs are frequently kept in pasteboard boxes with glass covers of proper size. Egg collections should not be exposed to daylight more than necessary, as they fade rapidly. Nests that are lined with hair or feathers are subject to attacks of insect pests and must be fumigated and cared for in the same way as skins.

CHAPTER V

COLLECTING AND PRESERVING AMPHIBIANS AND REPTILES

FRANCIS R. COOK

INTRODUCTION

Reptiles (snakes, lizards, turtles) and amphibians (frogs, toads, salamanders, newts) form an interesting but little-studied group of vertebrate animals in Canada. These two classes are often referred to together as *herptiles* or *herpetozoa*, and their study is called *herpetology*.

In Canada, 81 species of herptiles have been recorded: 42 reptiles and 39 amphibians. Sixty-three have only one race (subspecies) in Canada, and each of the remaining 18 is represented by two or more subspecies. The total number of forms, obtained by adding all the subspecies and those species with only one form, is 106. A few additional forms may be added as the amount and thoroughness of collecting increases. However, it is probable that no species new to science remains undiscovered within Canada as none of the species currently recognized are restricted to this country, and most are more extensive in range, and often more abundant, south of our border. Further studies of variations within species will likely reduce the number of valid subspecies.

Collections of Canadian herptiles, although not adding to the description of new taxa, are needed to further studies in variation, distribution, and life history. Detailed information about a species' distribution is basic for determination of the factors which restrict it, and museum specimens whose identity may be verified are the only sound basis for such information. Studies of variation within a species should be compiled from series of at least 20 specimens of each size and age class per locality taken from throughout the species range. Such studies often reveal differences between geographically separated populations which provide, even when the differences are not sufficient to warrant subspecific status for these populations, evidence important in zoogeographical and ecological studies. The number of series sufficient to give a true picture of variations within any one species can only be determined by the amount and distribution of the variations. Similarly, variations in life history may be determined from series. In other studies, involving only field observations or laboratory experiments, a few specimens should also be preserved so that the species or race to which the study pertains may be verified.

The National Museum of Canada needs additional material for its herptile collection to supplement that collected in its own systematic surveys of Canadian species. Further information is given under *Shipping*.

Many guide books provide notes on collecting and preserving reptiles and amphibians. The purpose of this chapter is to provide a more complete summary of the different techniques which have been used, together with an extensive bibliography of original papers on these subjects. A selected list of references provides the important handbooks and general texts necessary to identify Canadian species and to provide background information for their study. As yet there is no comprehensive book on the reptiles and amphibians of Canada, but one is in preparation.

COLLECTING

Legal Restrictions

In most areas there are no restrictions on collecting amphibians and reptiles, although official permission must be obtained to collect in national and provincial parks, and trespassing without permission on private property should be avoided.

Collecting Containers

It is usually most convenient to collect reptiles and amphibians alive. Small specimens, particularly salamanders and small frogs, are best placed in a jar when collected. Usually even a jar with a tight lid will serve for a short period (several hours, or longer) as these animals do not use the limited oxygen at a rapid rate. For longer periods the lid may be ventilated by punching holes (from the inside out) in it, or by cutting out a section and inserting a circle of wire screening which is held in place between the edge of the jar and a rim left around the top of the lid. A small amount of water should be added for moisture, but not so much that specimens cannot easily hold their heads above it. Moss, leaves, and the like may be added but are not necessary.

Cloth bags, such as flour, salt, and sugar sacks, are ideal for snakes, turtles, lizards, and larger frogs. They must be restitched to ensure strength of the seams and may be provided with a drawstring top if desired. These can be carried looped through one's belt. If no drawstring has been provided, the top of the bag may be knotted. With a drawstring, there must be enough excess string so that it may be wound around the neck of the bag and tied securely. For amphibians the bags must be thoroughly soaked to prevent desiccation of their contents. Bags of heavy plastic are recommended by some collectors. Canvas bags are too heavy, and the weave of burlap is too loose to make it a reliable container.

Care must always be taken not to leave collecting jars or bags in the direct sun, as the rise in temperature in the containers will quickly kill specimens. Both bags and bottles should be thoroughly washed before re-using.

General Collecting

Reptiles and amphibians may be encountered in a wide variety of natural environments—woods, fields, ponds, marshes, lakes, rivers, and streams. They may be terrestrial, arboreal, fossorial, or aquatic. Many species, especially those most successful in Canada, are found in a variety of habitats, but others are quite specialized in habitat and restricted in distribution. To collect the latter, a good text which gives what is known about the habitat preferences of the species must be consulted to determine the most likely places to search for it in a given area.

Often the discovery of specimens is purely a matter of chance, and the somewhat flippant but time-honoured reply to the question of how many specimens a good collector may expect to obtain on an ideal collecting day, 'none or more,' is all too true. The collector should always be prepared to collect a specimen of a rare species when he sees it, rather than assume he will get a second chance at a more convenient time.

The simplest technique for collecting most reptiles and amphibians is simply to slap a hand on or over them. Snakes may be pinned with a foot or a stick and picked up just behind the head. When held in this manner, a snake should also have a hand placed under it to support the body or it may thrash violently enough to snap its neck. The inexperienced person *never* should pick up or handle poisonous snakes. (In Canada the only poisonous snakes known to occur are rattlesnakes. These may usually be recognized by the rattles on the tail, but an occasional specimen may have its rattles broken off.) When in a rattlesnake area there is no substitute for being able to identify the local snakes on sight. Non-poisonous snakes have many fine needle-sharp recurved teeth, and the larger ones may inflict a painful bite. However, this is not dangerous unless the wound becomes infected. Frogs should be held around the body just in front of the hind legs. Lizards and salamanders may be held behind the head or by the body but never by the tail. In some species the tail readily snaps off. Frogs and salamanders are incapable of inflicting wounds, and even the bite of our larger lizards is usually no more than a painful pinch. Turtles should be grasped with one hand on either side of the shell, by the rear edge of the shell, or by the tail. Particular care must be exercised when handling Snapping Turtles (*Chelydra serpentina*) and Softshelled Turtles (*Trionyx spinifer*) as both can inflict nasty wounds with their razor-sharp jaws. Their long necks allow them to reach some distance back over their shells. Most other turtles may give a painful nip but rarely break the skin. Because of their sharp claws, the flailing feet of a struggling turtle are to be avoided.

Collecting Techniques

Permanent and temporary water. Many reptiles and amphibian species are associated, at least for some period in their life history, with permanent or temporary water habitats. Some species are restricted to this environment. The collector should walk around the margins of all water areas, alertly watching for specimens. Frogs, snakes, and turtles may all be found basking on the edge of such areas or on logs, boards, vegetation, and debris which project above the surface of the water. For larger ponds, rivers, and lakes, a boat may prove very useful as it allows the collector to approach from the open water side of a resting animal. It is essential to approach such areas carefully and make as little disturbance as possible.

Both day (particularly a warm, sunny one) and night collecting are profitable. A dip net is very useful, and hook and line, seines, and traps can yield specimens difficult to obtain by hand. These are discussed below. A good pair of binoculars (8 x 30 or 7 x 35) is invaluable to carefully scan for animals from some distance and to locate and identify specimens that might otherwise slip away unnoticed.

Some special methods for collecting turtles from a boat have been discussed by Haney and Smith (1950). Water goggling has been recommended by Marchand (1945) as effective in both observing and collecting aquatic turtles. Svihla (1959) has given a method of collecting the specialized stream-dwelling tadpoles of the Tailed Toad (*Ascaphus truei*). Martof (1963) has discussed a damming procedure for diverting one channel of a two-channelled stream for effective collecting in this habitat.

Cover. One cardinal rule in herpetological collecting is that the competent collector literally 'leaves no stone unturned' in his search for specimens.

Most species may at least occasionally be found under loose stones, boards, logs, bark, and similar cover. This is equally true in both terrestrial and aquatic habitats. Some species are rarely seen unless suitable cover is looked under. In places where little cover is available, good results may sometimes be obtained by placing boards, tar paper, and similar objects on the ground. Subsequent inspection of these will yield specimens. Care should always be taken to replace disturbed objects to their original position so that they will continue to offer suitable retreats for herptiles. Turtles under cover of vegetation or mud on the bottom of a pond may be detected by feeling for them with the hands or bare feet (*see* Lagler, 1943, p. 22). On land they may be discovered in piles of leaves and similar cover by 'sounding' with a wooden pole as described by Carpenter (1955).

Amphibian breeding aggregations. Many amphibians appear at temporary ponds during the first warm days after the snow has melted. The earliest breeders may even start calling before the ice on the ponds has completely melted if daytime air temperatures have been warm enough. The most vigorous calling and breeding activity is usually at night. For some frogs and salamanders the breeding period is remarkably short—as little as two weeks elapsing between the arrival of the first individual and the departure of the last if the weather is consistently warm. Breeding frogs form large choruses which can be heard from some distance. The salamanders which breed in the same temporary ponds usually arrive at the same time as the first frogs are calling. The early breeding frogs are followed by other species as the weather gradually becomes warmer, and usually several species of frogs may be heard on the same night. The latest frogs to breed are those which lay their eggs in permanent water. These do not start to call in appreciable numbers until late spring or early summer.

In the grassland regions of the prairie provinces and British Columbia, many amphibians breed only during or after heavy rains in the late spring or early summer. They are difficult, often impossible, to find except during this brief (often only one or two nights) period. In exceptional years when no heavy rains fall during this time, they may not breed at all.

Night is the best time to collect breeding amphibians, and a headlamp or flashlight (*see* below) is essential. Most of these species are difficult or impossible to approach in the daytime, and some are active only after dark.

Other amphibian aggregations. Amphibian eggs are usually laid in large numbers, and often the percentage of larvae which develop to transformation is high. The developmental rate of species that lay in temporary ponds is rapid, so that the first transforming frogs are ready to leave the pond by early summer. Often large aggregations of tadpoles, salamander larvae, or transforming individuals can be found at or near a pond margin in the open or under suitable cover. Recently transformed individuals of one species or another may be found at the margin of aquatic habitats throughout the summer and early fall. Often species that were overlooked during their breeding season may be identified from larvae or transforming individuals.

Hibernating dens. In the spring or fall, snake dens may occasionally be located. During the first warm days of spring, snakes emerge from such hibernating sites to sun themselves at or near the entrance. Dens are often in rocky outcrops on south-facing slopes where there is access to a dry location below frost penetration. Often one den serves as winter quarters for hundreds of snakes of several species from a large surrounding area. Not all species, nor perhaps all age groups of a species, use such sites. Individual snakes and small groups of snakes may winter in rodent burrows, small crevices, and even in ant hills. During the summer the snakes disperse, and few remain in a den area. In the fall during the last warm days before winter, they again gather at the den and sun themselves at or near the den entrance. Dens are best located by questioning local residents who may remember a place where they have seen large concentrations of snakes in the spring or fall and may be able to give directions to it. Migrations of snakes across roads in the spring and fall are additional evidence of the proximity and direction of a den. A classic study of a snake den has been presented by Woodbury, *et al.* (1951), and additional comments and bibliography are given by Woodbury and Parker (1956).

Road driving. Good collecting results may be obtained by driving along rural roads at 10 to 15 miles an hour and stopping to collect or identify each specimen seen. Often dead specimens will be fresh and sufficiently intact to be worth preserving. A record should be kept of every specimen seen, whether collected or not, and localities may be noted by mileage readings, to a tenth of a mile, from the nearest town along the road. On well-travelled roads, where slow driving and repeated stops are illegal and dangerous, a white cloth attached to a weight might be tossed to the side of the road to mark the spot where a specimen was seen until the car can be turned around at a more convenient place. Road driving is particularly effective for snakes in some areas, and Schmidt and Davis (1941), Fitch (1949), Campbell (1956), and others have presented examples of the data which may be obtained. Early morning, late afternoon, and after dark are the most productive periods. Rainy nights, particularly warm nights in the early spring, are the most productive for amphibians which migrate to breeding ponds under such conditions. Female turtles may be found crossing roads in the late spring and early summer when they are searching for suitable egg-laying sites.

Collecting Equipment

Snake sticks and grippers. Various devices have been invented for use in capturing and restraining snakes. A stick, forked at the tip, has been used to pin snakes, but it is ineffective unless the ground beneath the snake is soft. A stick with an 'L' metal bracket at the end is useful to pin snakes or to slip under them to lift them up. A stick with a 'J'-shaped metal end is also used for the latter purpose. Most snakes when lifted by mid-body with such a stick will balance themselves on it when held off the ground. Snake grippers or snake-tongs are an excellent device for capturing or holding snakes. They are made of aluminium, with simple jaws at one end and a handle with a lever to close the jaws at the other. They have been described and figured by Pillstrom (1954) and may be obtained from a commercial manufacturer.

Potato hoe and stevedore hook. The three- or five-pronged potato hoe, obtainable at hardware stores, is very useful for turning boards, logs, stones, and other cover when searching for specimens. The stevedore hook is similarly used and has a 'T'-shaped handle and a 'J'-shaped end for gripping objects to turn them. Both reduce the labour involved in turning cover and are essential where such cover may shelter poisonous snakes.

Headlamp. A light which can be strapped on the forehead with a container for batteries or a battery pack that can be attached to the belt or carried in a pocket is essential for night collecting. A strong flashlight may be used; however it has the disadvantage of being carried by hand. A headlamp leaves both hands free to capture specimens and place them in containers. Headlamps may be obtained at hardware and sporting goods stores. Most amphibians and reptiles are not disturbed by artificial light and may easily be approached and netted or grabbed by hand. Frogs will usually continue to call even when the beam of the light is directly on them. Care has to be taken to approach them with as little disturbance as possible however, as both noise and surface waves on water will sometimes cause them to dive for cover.

Dip nets. These may be purchased from biological supply houses or made at home. A rust-proof metal hoop is fixed to a wooden pole, and a net is sewed onto the hoop. The edge around the hoop should be covered with a strip of cloth. The netting should be sufficiently sturdy to withstand rough usage, and the mesh should be fine enough to retain small tadpoles and salamander larvae. A net which is wider at the bottom than at the hoop and fairly deep is the most effective kind. This hinders the more robust frogs from jumping straight out.

Nets of larger hoop diameter, coarser mesh, and longer pole length may be made for turtles.

Seine. A seine is often effective for aquatic adults and larvae. It requires two people to operate a seine effectively. Care must be taken to keep the lower edge on the bottom to prevent the escape of animals beneath it. In small creeks and streams it should be fixed across the channel; with it in place the collectors then move towards it from a point upstream, turning over every stone and other loose object in their path. Specimens and debris will be swept downstream into the seine by the current. A seine is not effective in areas of heavy vegetation because the bottom tends to roll up when dragged through rooted plants. For such situations, devices described by Strawn (1954) and Goin (1942) may be the most practical.

Electric shocker. Anderson and Smith (1950) and Gunning and Lewis (1957) describe electrical shockers that are said to be effective for catching amphibians and reptiles.

Hook and line. Many of the larger aquatic species can be taken with a hook and line. Large adult and larval salamanders and turtles may be caught occasionally with lines baited with meat or fish. Lagler (1943, p. 22) gives instructions for set-lines for turtles. A wire leader must be used. The mudpuppy (*Necturus maculosus*), an eastern aquatic salamander, is often taken in the winter by fishing through holes in the ice.

The larger frogs will take a hook with a piece of cloth attached to it. This should be cast in front of them and flicked to simulate a moving insect.

Guns. In some situations it is impossible to approach specimens closely enough to catch them by net or by hand. A .22 pistol or rifle, loaded with .22 dust shot, is effective for frogs, lizards, and snakes. The barrel should be bored smooth to obtain the full benefit of the shot pattern. For turtles, a .410 shotgun must be used. Care must be taken to shoot at close enough range to kill the specimen but not so near that it will be severely damaged. This method is especially recommended for rattlesnakes but should be used for other species only as a last resort, because the likelihood of damaging specimens is great. For areas where the use of a gun is prohibited, other devices have been used for collecting lizards and frogs. Shot may be fired from a slingshot or a wooden 'gun' (Neill, 1956). Alternatively a chain or rubber band may be used to stun specimens (*see* Brown, 1946; Dundee, 1950).

Snares. Various types of hand-snares have been used effectively for reptiles, particularly lizards. These animals often cannot be approached closely enough to grab by hand and are often in situations where a net is impracticable. They may be noosed with a loop of wire, usually on a pole, as they will sit quietly until the loop is placed over the head and pulled tightly around the neck with a sudden jerk. Eakin (1957) has described such a device, and Stickel (1944) has presented a model in which the loop is closed instantaneously by a trigger mechanism. Franklin (1947) describes a type of snare for water snakes.

Pitfall traps. Simple but effective traps for both reptiles and amphibians have been devised on the pitfall principle. A container (glass jar or tin can) is sunk into the ground so that its top is flush with the surface. Several inches of water are placed in a shallow-trap to prevent the animal from jumping out.

Banta (1957) describes a trap of this type for lizards in which a five-quart tin can was used (height 240 mm, inside diameter 66 mm). A disassembled cardboard carton was used as a cover, with small rocks under it to hold it above the surface of the ground and with rocks on top of it to keep it in place. The animals seek cover under the cardboard and fall into the trap. Rogers (1939) gives the plans for a plywood pitfall trap which has a balanced cardboard cover over part of the top. This tilts when an animal runs over it, and it drops into the trap. Lannom (1962) successfully tried a lizard pitfall trap which had artificial trout flies suspended from a short thread above it. Lizards leaping for the fly would fall into the trap. Breen (1949) has described an effective turtle trap based on the pitfall principle. A barrel is weighted with stones and sunk in the water so that its top is level with the water surface. A board attached to the edge and partly submerged in the water acts as a ramp, and a piece of raw meat is placed on it at the end over the barrel mouth. A turtle ascending the ramp is plunged into the barrel after it passes the point of balance. Another type of turtle pitfall (Lagler, 1943) is made by constructing a rectangular wooden frame with a wire hamper attached below it. Spikes or large nails are driven in along the inner edge of the frame so that they project downward, and their heads are clipped off. The trap floats in the water, and turtles will climb up on the wooden frame to sun themselves. If they leave by plunging into the centre of the trap, the projecting spikes will prevent them from climbing back out. Trenches with smooth steep sides, dug for construction or laying pipe, often inadvertently serve as pitfall traps for herptiles.

In using pitfall traps, the amount of success will depend on the placement of the traps and the amount of activity and abundance of the animals to be trapped. For amphibians, a rainy night will usually provide the largest catch, provided of course that the trap is placed so that it will not fill up with rainwater and release the catch. Traps should be inspected *at least* once a day and the captured animals removed. Pitfalls must be removed or filled in when they are no longer in use, otherwise they will become an unnecessary graveyard for a wide variety of animals from small mammals to insects.

Funnel traps. Various types of traps involving a funnel which allows an animal easy access to a trap but prevents its exit have been employed for herptiles.

Fitch (1951) presented details for an effective simple funnel trap for catching snakes and lizards:

> "The model used consists of a piece of hardware cloth wire, one-fourth or one-eighth inch mesh, rolled into a cylinder and held in this shape by having the edges turned back and pounded together. An entrance funnel of the same material is fitted firmly into each end. First, each end of the cylinder is turned inward at right angles, for half an inch or so, forming a skirt. The elasticity of the hardware cloth tends to hold the funnel in place when it has been forced into the cylinder as far as it will go. Shingle nails woven through the meshes of the funnel and the end of the cylinder to maintain firm contact between them provided reinforcement which was found to be especially desirable in traps liable to be disturbed by predators. Striped skunks, spotted skunks, raccoons and probably domestic cats occasionally broke open the funnel traps to prey upon the trapped animals. Effectiveness of the traps is increased by attaching a valve-type, transparent, cellulose acetate door inside each funnel opening. This permits use of a larger entrance through the funnel. The door pivots on its upper edge, which is perforated and threaded with a fine wire attached to the end of the funnel. A trapped animal can be removed easily by putting the funnel from one end of the trap and shaking the animal into a cloth bag."

Fitch used two sizes of traps, 7 inches long and 3 inches in diameter for small lizards, and 15 inches long and 6 inches in diameter or even larger for snakes and larger lizards. The traps must be sheltered from direct sunlight by setting them in the woods or by placing rocks or boards over them. In this study traps were placed where natural objects such as sunken logs, walls, or rock outcrops would guide the animal to the mouth of the funnel.

Imler (1945) and Dargan and Stickel (1949) have described slightly more elaborate funnel traps for snakes. The latter authors used funnel traps with drift fences of hardware cloth, 12 inches high, which were extended 25 feet from each end of the trap. A 2-foot wing was placed obliquely at each corner of the trap to help guide snakes into the funnels. Snakes followed the drift fences to the trap.

Another model of funnel trap, used for lizards, was described by Vogt (1941). This consisted of a wooden platform, one metre square, raised 5 cm off the earth by narrow boards nailed to the edge of the underside. Two gaps were left in diagonally opposite corners, one closed by a plug and the other containing a funnel. The bottom was wire mesh. The attraction is the cover provided by the board. Animals may be taken out by removing the plug and shaking the trap. In these traps no bait was used although Jorgensen and Orton (1961) give evidence of some lizards having eaten oatmeal in traps.

Moulton (1954) used a rectangular trap of wire screening on a wooden frame with a funnel inserted in an oval opening $2\frac{1}{2}$ inches above the floor of the trap. The outside entrance of the funnel was made level with the ground by placing the trap in a shallow depression. A collar was placed around the inner end of the funnel as an aid in preventing the escape of captured animals. The funnel of this trap was placed in an opening in a low fence of tar paper, reinforced with fine wire and held upright by small stakes driven into the ground at intervals on both sides of the fence. The fence covered a portion of the periphery of a pond to which spring breeding amphibians were migrating, and the arrangement was effective in trapping them on this journey. Storm and Pimentel (1954) also employed a fence and funnel traps to collect amphibians on their way to a breeding pond.

Carpenter (1953) employed the Fitch model funnel trap for aquatic amphibians. The traps which had a 6- to 10-foot rope attached were cast into the water the length of the rope and pulled into a position where the funnel openings were parallel to the shore line; the rope was attached to an object on the shore. Frogs and adult and larval salamanders were taken. The writer has had success in trapping both newts and salamander larvae with standard commercial minnow traps. The traps may be set unbaited, or raw meat or fish may be used as bait.

For turtles, various models of funnel traps, usually called 'hoop traps,' have been designed and found to be very effective.

Legler (1960) describes an excellent and simply constructed model:

"Hoops are made from aluminium tubing. Tubing with a diameter of $\frac{1}{4}$ inch and walls approximately $\frac{1}{16}$ inch thick has the most advantageous combination of light weight and necessary strength for hoops up to two feet in diameter. Heavier tubing must be used for larger hoops. Tubing can be bent into hoops by hand, using the top of a circular object (crock, paper basket, or any other round object of proper size) as a form. The form used should be several inches smaller than the desired diameter of the hoops, inasmuch as the inherent springiness of the tubing causes it to expand considerably after it is bent. Mechanical devices for bending tubing should be avoided because they tend to flatten the tubing. The open ends of the hoop are joined by a two-inch sleeve, cut from a piece of tubing with an inside diameter of $\frac{1}{4}$ inch. The sleeve can be crimped with pliers to insure a tight fit.

"The body of the trap, or bag, consists of a single piece of commercially made netting. Netting of $\frac{3}{4}$ inch mesh (by square measurement, i.e., measured from knot to knot), made from No. 12 cotton twine, is sufficiently small and strong to retain most animals that will enter the trap. Four hoops are threaded into the rectangle of netting of the proper size (large enough to fit snugly around the hoops, to allow the hoops to be spaced at twelve-mesh intervals, and to allow for a throat 12 meshes deep on each end) and the free edges of the netting are woven together by a single piece of twine stretched between the end hoops; this seam is then reinforced by knots on alternate meshes. The free edges of the throats are similarly closed with knots, puckered to the desired size with a single piece of twine and reflected into the body of the trap. Netting should be treated with a preservative to prevent rotting from repeated wettings. It is most practical to purchase netting that has been treated and dried at the factory (copper napthanate is the substance most often used), the extra cost of this service is negligible when one considers the trouble involved in soaking large pieces of netting in a small workshop or laboratory. Twine used to join pieces of netting must also be treated with preservative.

"Nets 19 inches in diameter, 33 inches long, and with a throat 12 inches deep on each end were found to be the most useful in collecting turtles. Nets as small as 12 inches in diameter are of limited use and are expensive to make. Nets as large as four feet in diameter are useful if provision can be made to fit them (and their necessarily long stiffeners) into the small spaces ordinarily available for field gear. The opening of the throat in my nets is approximately 10 inches wide and 3 inches high when the nets are stretched into position for setting, but size of throat-opening can be varied simply by adjusting the length of the cord passing around the opening. It should be borne in mind that the width of the throat-opening will determine the maximum size of turtle that can enter the trap. The vertical diameter of the throat-opening expands as a turtle enters the trap; seemingly turtles are little hindered by having to push through this low opening. The throats are held open by two lines tied to the second hoop of the opposite end; lines from the two throats cross each other at the sides of the trap between the two centre hoops.

"Traps are held open and rigid by stiffeners made from $\frac{1}{2}$ inch wooden doweling; metal screw-hooks on the ends of the stiffeners hook into the end hoops and hold the stiffeners in place. A useful modification of the stiffener described is to anchor a screw-hook at one end and to solder the other screw-hook to a copper sleeve fitting loosely over the wooden rod; this produces a friction sleeve which is held in place as long as tension is exerted on the screw-hook and permits use of the same size of stiffener for traps that vary slightly in length (due either to shrinkage or to inconsistency of construction). Wooden stiffeners must be coated with paint, varnish, or lacquer to prevent soaking and subsequent warping.

"A bait-holder, made from a square of $\frac{1}{4}$ inch mesh hardware cloth is suspended from the inside of the net so that it hangs between the openings of the throats and just above them. The bait-holder is simply folded around the bait.

"The nets herein described can be set effectively in any suitable aquatic habitat having water deep enough (at least 10 inches) to cover the throats. My experience has been that traps are most effective when set in shallow water near shore, in snags, or beneath undercut banks parallel to objects along which aquatic animals are likely to be moving. If traps are not checked at frequent intervals turtles caught in them will drown.

"Any kind of fresh (juicy or bloody) meat or fish will generally make good bait for turtles. . . . Canned sardines (in oil) can be purchased almost anywhere that food is sold and make an ideal general purpose bait when conditions are such that maintaining fresh bait is impossible. A trap baited with one or two sardines will frequently catch turtles and will nearly always catch fish, which can in turn be used as fresh bait."

FIELD NOTES

The value of time and effort invested in collecting, measuring, and preserving specimens is always enhanced if careful and complete field notes have been recorded. Even without the specimens, field notes are valuable although often the original collections are necessary to couple observations with the correct species or subspecies to which they refer. Each collector usually develops an individual system which suits his particular purpose. However, two principles should always be kept in mind: (1) the field notes may be of assistance to future workers and therefore should be as clear and accurate as possible, and (2) all *data* and observations should be included, as information which may not appear useful at the time may prove valuable later, either in itself or as a clue to a productive approach in future studies. It is also essential to record all field notes at the time the observations are made. A pocket notebook, preferably with a hard cover, is best for such notes.

The following outline should prove useful as a guide to important categories and a method or arrangement. A new page is used for each locality.

1. *Date.* Day, month (*always* written out, never expressed as a number), and year.
2. *Locality.* Always record in miles and direction from the nearest settlement with a Post Office.
3. *Collector(s).* All collectors should be listed. It is customary to place the name of the field note compiler first.
4. *Time.* Hours and duration of collecting.
5. *Weather.* Include both general description and exact temperatures (if taken).
6. *Habitat.* Describe the topography, elevation, and vegetation (deciduous forest, open fields, etc.). Where plant and other animal species are known, list them and their relative abundance. (If practicable, collect specimens to be identified later. This can be of particular importance where visible animals may be food items that may be compared with stomach contents of herptiles collected.) If specific identification of plants is not possible, at least the general growth type of the dominant plants should be given (e.g., tree, bush, grass, for terrestrial plants; and submergent, emergent, floating, for aquatic plants) and any general identifications (e.g., spruce, oak, cat-tail, water-lily). The extent of the area covered should be noted. Always write from the general to the specific.
7. *Species of Herptiles.* Include both those collected and those observed only. Under each species an estimate of numbers should be made. Such terms as 'abundant,' 'infrequent,' 'rare' may be used, but actual counts or estimates of individuals are more meaningful. The microhabitat (e.g., pond edge, under spruce log, etc.), and behaviour and colour and size variations should also be noted for each species.

The pocket notebook may be used either as an outline for entries in a larger field journal or as the finished record. Even in the former case it should always be kept as a check on errors in recopying. If a longer journal is used, it may also include pages for use as a catalogue of collections and a record of measurements of specimens. Maps and itineraries of collecting localities should always be kept. All collecting localities should be marked on the maps. Photographs of collecting areas and specimens in the field are invaluable.

PRESERVING

Herptiles are usually brought back alive to the laboratory or home, and preserved there. To obtain the best results the following steps should be taken:

1. Colour notes and photographs.
2. Killing.
3. Measuring.
4. Labelling and cataloguing.
5. Preservation:

Adults in 10 per cent formalin, injected, fixed, checked, and stored.

These headings are discussed in detail below:

Colour Notes and Photographs

In amphibians, some colour change often occurs at death. All reptiles and amphibians preserved in formalin and alcohol lose colour over a period of time. If possible, therefore, detailed colour notes should be taken before a specimen is killed. Colour photographs of living specimens should also be taken when practicable.

Killing

It is important to kill specimens so that, relaxed after death, they may be easily measured and arranged uncontorted in preservative. Direct immersion in formalin or strong alcohol results in stiff contorted specimens since they die in severe pain.

Ether is excellent for killing specimens if used carefully. A small amount is poured in with specimens confined in a glass jar which has a tight lid. Care must be taken not to use too much or specimens will stiffen. Fifteen to thirty minutes will kill most specimens. If too little is used, or if specimens are left in the jar an insufficient length of time, they will revive.

Chloroform is effective for killing small and medium-sized turtles and may be used in the manner described above. It will kill other herptiles, but they almost always die stiff and contorted.

Warm water (110°–120° F) will kill both reptiles and amphibians. They may be enclosed in a cloth bag and immersed in the water. They must be removed immediately after death, or they will become rigid in the position in which they died.

'Nembutal,' the commercial name for Pentobarbital Sodium, can be used to kill reptiles by injecting a dilute solution into or near the heart. The aqueous veterinarian Nembutal should be used, *not* the syrupy elixir. The commercial Nembutal should be diluted with nine parts of water for each part of Nembutal. Prepare only small quantities as the solution soon loses its strength. Small lizards and snakes require only a few drops, but large snakes (over two feet) and turtles will require one cc or more.

'Chlorotone,' the commercial name for Hydrous Chlorobutanal, may be used for killing amphibians by drowning them in it. A few crystals to a quart of water makes a suitable killing solution.

Ethyl alcohol, diluted to 10 per cent may also be used in the same manner. Specimens should be removed as soon as they die.

Procaine Hydrochloride may be injected into reptiles and amphibians to kill them (Livezey, 1958). A 10 per cent solution is made by dissolving tablets of the drug in water. Each tablet contains 0.07 gm. It may also be obtained in crystalline form or in a prepared 1 and 2 per cent solution. Most reptiles will be killed by an injection near the heart of 0.05 cc (=0.47 mgm) per gram of body weight. Amphibians may be killed by drowning in the solution as well as by injection.

Measurements

Although specimens are not usually measured before preservation in herpetological studies, this procedure is strongly urged here whenever practicable. Too often inaccurate measurements are obtained from specimens which have been preserved for varying lengths of time. This is due to distortion and/or shrinkage of specimens in preservative. Careful measurements of fresh, relaxed specimens, taken immediately after they have been killed, are infinitely more valuable. All measurements should be recorded in millimetres. The length of most Canadian frogs, salamanders, and small lizards may be taken with a 152-mm rule. Snakes, large lizards, and very large frogs and salamanders will require a tape measure. Leg length (tibia) for frogs may be taken with hand calipers. All measurements should be at least estimated to the nearest half millimetre.

The following should be recorded:

Snakes and Lizards and Salamanders—

(1) Body length—taken from the tip of the snout to the vent (posterior angle of vent in salamanders) along the underside of the body.

(2) Total length—taken from the tip of the snout to the tip of the tail.

Frogs—

(1) Body length—taken from tip of snout to centre of the vent by placing the ruler along the midline of the back.

(2) Tibia length—taken from 'knee' to 'heel' of the hind foot when it is flexed.

Turtles—

Carapace and plastron length and width, taken at widest portions. (This is a point-to-point straight line measurement with calipers—never along the curve of the shell.)

Measurements may also be taken of the length of the head and neck—with the neck extended—from the tip of the snout to the junction of neck and body, and the length of the tail—from the tip of the tail to the vent, and from the tip of the tail to the junction of the tail and body.

Labelling and Cataloguing

Labels should be of high (preferably 100%) rag-content paper which takes ink well. (The Dennison Paper Company, Drummondville, Quebec, manufactures a suitable paper for labels: Resistall Index Bristol, 100% Rag, 110 lb. wt.) The writing should be in waterproof india ink or soft pencil.

Each group of specimens of one species having identical collecting data should be kept separate from all other collections and should have a label giving at least *locality* (miles and direction from nearest settlement with a post office), *date* (e.g., 12 June 1964, *always* write out month in letters and give all four digits of the year), and *collector(s)*. Usually a collector will have his own series of catalogue numbers, which will also be placed on each label. These may run consecutively through his lifetime, or he may start a new series each year prefixed by the last two digits of that year. The collectors initials should always prefix numbers in his series, e.g., FRC-1 or FRC-64-1.

Specimens may each receive an individual catalogue number, or they may be catalogued by lot, giving the same number to all specimens of one species collected on the same day, in the same place, by the same collectors. If catalogued individually, each specimen should have an individual label giving full data and catalogue number. If catalogued by lot, each specimen should have a label with the catalogue number of the lot to which it belongs, and an individual sub-number within that lot which is circled (e.g., FRC-64-1 ①, FRC-64-1 ② etc.). Individual labels should be tied to the specimen with strong thread. Specimens in the same lot should always be kept together in one container in which there is a label giving full data for the lot. Two lots should *never* be mixed in the same container. A catalogue book or separate pages in the field notebook should be kept giving each number and the essential data. Separate pages should list the measurements for each specimen and its full catalogue number and data.

Additional data such as the scientific name of the species (if known), habitat, and other collection information and measurements may be recorded on the back of the label, if desired.

Preservatives

Herptiles are usually preserved whole in a liquid preservative. Although there are several suitable preservatives, formalin is the most satisfactory for initial preservation. Commercial formalin is a solution containing approximately 37 per cent formaldehyde gas dissolved in water. Clear, rather than amber formalin should be used, and it is readily obtainable at most drugstores. It hardens the tissues of specimens and immediately prevents decay. For the purpose of preserving specimens, commercial formalin is considered as a 100 per cent solution. A 10 per cent solution is best for preserving specimens (1 part formalin to 9 parts water). If particular care is taken with injecting and checking specimens, a weaker solution—5 per cent—can be used as an initial preservative for snakes, lizards, and amphibians, and these may be changed after 24 hours into fresh 5 per cent for snakes and lizards, and 3 per cent for amphibians. Turtles must always be preserved in 10 per cent. The disadvantage with formalin is its strong, irritating odour. Some people develop a strong allergy to it.

Although formalin is usually purchased as a liquid, a solid polymer of formaldehyde, *paraformaldehyde*, is available. This has transportation advantages in weight reduction, reduced danger of leakage, and ease of shipment (Taub, 1962). An efficient method of preparing a 10 per cent formalin solution from it has been outlined by Huheey (1963): 16 grams of paraformaldehyde and 4 grams of anhydrous sodium carbonate are mixed. A small amount of a wetting agent (e.g., Alconox) appears to disperse the solid more readily and change the solution. The dry mixture may be kept in glass vials or in pockets made of polyvinyl film sealed with a hot iron. The contents of one pack dissolved in 400 ml of cold water yields a buffered 10 per cent formalin solution.

If formalin is not available or cannot be used, 95 per cent (reptiles) or 70 per cent (amphibians) ethyl or 40 per cent isopropyl alcohol may be used as the initial preservative. Specimens initially preserved in these solutions have to be carefully inspected periodically during the first few days of preservation to watch for areas which have not preserved well. If preserved in ethyl they should be transferred to 70 per cent after 24 to 48 hours. As a last resort methyl, wood, or rubbing alcohol (add about one-fifth water) or strong alcoholic spirits (preferably 'overproof') may be used. Liquor which is 100 proof is only 50 per cent ethyl alcohol. If no liquid preservatives are available, specimens may be placed in a strong brine solution (one pound of salt to one gallon of water). If the specimens are intended for a museum and if weak alcohol or salt had to be used, these specimens should be shipped as soon as possible so that they can receive additional attention.

Specimens may also be frozen. They may either be thawed and preserved, as one would a fresh specimen, or placed in a solution of 10 per cent formalin to thaw and injected later.

The following procedure is best for most adult and juvenile specimens. Large specimens, tadpoles, larvae, and eggs are discussed separately, as are special methods which retain skin colour.

Injecting. All herptiles, except very small frogs and salamanders, should be injected with preservative using a hypodermic syringe. A 50 cc capacity syringe is the most convenient. Snakes should be injected in several places throughout their length; frogs, lizards, and salamanders need only a single injection in the body cavity. Turtles should be injected in the body near the insertion of each leg, and the legs and neck should also be individually injected so that they stand out from the body. If no syringe is available, a single slit may be made with a sharp scalpel or scissors to one side of the midline of the underside in large frogs, salamanders, and lizards; several slits of an inch or more in length and two or three inches apart may be made along the underside in snakes, and in the legs and flesh parts between the shell in turtles. Often the best results with snakes are obtained by a combination of both injection and slitting. A slit should always be made along one side of the underside of the tail. In male snakes and lizards, one hemipenis should be everted by pressing at the base of the tail, and the specimen should be preserved with the hemipenis everted. An injection of preservative into the base of the tail will keep it extended.

A piece of absorbent cotton, large enough to hold the jaws open, should be inserted into the mouth so that it may be readily examined without undue damage to the specimen after it has hardened in preservative. If the cotton is placed in the mouth of a frog before measuring, it aligns the head with the body for measuring.

If reptiles and large amphibians are not injected or slit, they will often develop soft areas due to decay which began before the preservative was able to penetrate through the skin to those areas. Injection also serves to fill out the body in a natural shape. Over injecting, so that the body is unduly distended or bloated, should be avoided.

Fixing. After injection, specimens should be placed in a shallow tray or glass jar. Put at least enough formalin in the container to more than cover all individuals. Specimens should not be crowded in containers, especially at this stage. As specimens soon become hard and inflexible after a few hours in formalin, it is important at this time to arrange them in the desired positions. Snakes should be coiled in an oval shape; lizards and salamanders should be placed straight (although in large specimens the tail may be bent back along the body); frogs should be placed with hind legs folded at sides in a natural resting position; and turtles should have the head and neck, legs and tail extended from the body by the injection of formalin. For larger specimens such as turtles and some snakes that will not fit available glass containers, earthen crocks with tight-fitting lids, rust-proof metal containers, or metal containers lined with a bag of tough plastic may be used.

Checking. After 24 hours, a specimen should be rigid and without a soft or discoloured spot. Such a spot indicates spoilage, and if present, a cut should be made through it and extended a short distance to either side.

Storage. After 24 hours the original preservative should be poured off and discarded, and a fresh solution of 10 per cent formalin should be added. Specimens may be stored in any type of wide-mouthed glass jars which have tight-fitting lids. If specimens are to be stored in formalin for more than a few weeks, one level teaspoon of borax should be added for each half gallon of 10 per cent formalin to buffer the solution, as formalin will form small quantities of acid which will decalcify bones and soften specimens. Permanent storage in most museums is in 40 per cent isopropyl or 70 per cent ethyl alcohol, as formalin is irritating to work with. However, for private collections, formalin has the advantage of being inexpensive. Specimens may be transferred to isopropyl or ethyl alcohol within a couple of weeks after the initial preserving.

Large specimens. Large snakes and turtles present a problem, as suitable containers in which to preserve them may not be readily available.

Snakes. As the head alone (if comparatively undamaged) can usually be identified to species, in extreme circumstances it alone may be saved, preserved in formalin, as a record of a particular species occurrence in an area. Very large snakes may be skinned after the method of Etheridge (n.d.): Make a single long cut in the belly, just to one side of the midline, beginning about an inch in front of the anus. *Do not* cut through the anal plate. Work the skin loose from the body but do not attempt to remove the skin from the head or tail. Sever the body from the head and tail and roll the attached head, body skin, and tail loosely together and place in 10 per cent formalin.

Turtles. Large turtles may be opened by sawing through the bridge on each side which unites the carapace and the plastron (Figure 45). The neck should be severed, and the flesh removed from the legs, tail, neck, and head as far as possible, after which the whole specimen may be rubbed with borax or washed in water and submerged in brine to cure. MacMahon (1961) recommends freezing turtles which have started to decompose before cleaning the shells.

Tadpoles, larvae, and eggs. Tadpoles, salamander larvae, and reptile and amphibian eggs may be preserved by placing them directly into 3 per cent formalin. Stronger solutions may also be used, but they are likely to cause shrinkage. The solution should be changed after 24 hours except in amphibian eggs. Some collectors pour enough 100 per cent formalin into water containing amphibian larvae or eggs to make the solution approximately 3 per cent or slightly more. Reptile eggs, at least, should be measured before preserving. The development of frog's eggs and tadpoles may be staged according to the tables in Gosner (1960).

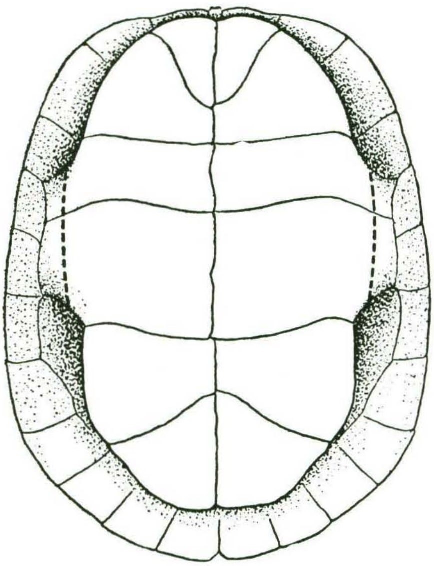

Figure 45. Shell of western painted turtle showing where cuts should be made to remove plastron or lower shell.

Preserving Skin Colour

Frogs. The pigments in the skins of amphibians are relatively unstable, and when the animals are placed in the more common preservatives such as formalin and alcohol, the colours, particularly the more delicate tints, are either entirely lost or greatly altered. The following method for preserving amphibian skin colours is from Kincaid (1948), as summarized by Anderson (1960) and modified from papers by Juszczyk (1952) and Turner (1959). The method is to skin the frog and mount the skin on a sheet of heavy paper or cardboard.

Frogs for this purpose are best killed with chloroform, as ether causes hemorrhage and resulting discoloration. The skin is slit along the middle of the underside from the anal area to the tip of the lower jaw, taking care not to cut the tissues beneath. Transverse cuts are made from the edge of the original cut across the underside of the forward portion of the body following the contour of the ventral side of the fore limbs, including the skin on the digits. Similar cuts are made to expose the under surfaces of the hind limbs with their digits. Very little effort is now required to loosen the skin from the underlying tissues. The contact with the anal area may require a snip of the scissors, and there may be some resistance when the skin is pulled over the head. The skin is next floated in a dish of tapwater, and any particles of adhering tissue are removed.

Remove the skin to a second dish of clear tapwater and float it out after the fashion of a piece of seaweed. Next, wet a piece of cardboard of suitable size and bring it beneath the floating skin with the pigmented surface uppermost. With a little care the cardboard can now be raised from the water bringing the skin with it and leaving it well spread out upon the surface. With a pair of dissecting needles, correct the position of the various parts and rearrange places where the edge of the skin has been turned under. With the finger or a pledget of cotton, smooth down the skin so as to remove all air bubbles. No adhesive of any kind is required.

The cardboard with its attached skin should now be put face up on a sheet of thick blotting paper to permit the greater portion of the moisture to escape and to allow the glutinous element in the skin to become firmly attached to the paper. As soon as the specimen has become partially dried out, but is still quite moist, place it between two pieces of heavy blotting paper or botanical driers, and put it under pressure until entirely dry. Any fibres from the blotting paper found adhering to the surface of the skin may be removed by rubbing gently with a moistened bit of muslin. Specimens preserved in this manner are reported to retain their natural colour for long periods. They must be stored away from strong light, and never exposed for any length of time to it.

Snakes. Beebe (1947) has recorded a method of preserving the colour of snakes' skins similar to that for frogs:

> ". . . skin [the snake] by a straight mid-ventral incision from neck to tail tip. Cut around the side of the head to the gape, so when the entire skull and jaws are free, the dorsal and ventral cephalic scales are left intact.
>
> "Have ready a sheet of moderately stiff cardboard.
>
> "Starting with the head, the skin is gently spread and pressed down evenly and firmly, working back slowly. At the bottom of the sheet it may be necessary to cut straight across the skin and begin again at the top. It is easier for two people to work together, one advancing with all eight finger-tips consolidating the contact, especially along the edges, while the other person spreads, flattens, and presses down. When the entire skin has been attached to the paper, it is placed upon any firm, flat surface, as a table, or the floor, covered with a sheet of blotting paper, on the top of which come newspapers or more cardboard, and firmly heavy weights, such as books. The blotting paper should be changed at least twice in two days, when the process is complete. Catalogue numbers, sex, locality, or any desired data can then be added, and the finished sheets with their skins filed in a plant press under moderate pressure to prevent any slight curling which may ensue.
>
> "The skinned body may be examined for sex, stomach contents, or embryos, labelled and preserved in alcohol [or formalin] as usual."

CARE IN CAPTIVITY

Care of Adults and Juveniles

Often it is desirable to retain a specimen alive for a short time before preserving it in order to note behaviour, mating, egg laying, or colour changes. Most amphibians and reptiles may be easily kept under artificial conditions in a terrarium or aquarium. For terrestrial species, a small dish of water and an object to crawl under are necessities. Aquatic individuals will do well in a bowl of water with some water plants, and most species need a small platform to allow them to crawl out of the water occasionally. Frogs and toads thrive on live insects; salamanders on small earthworms and whiteworms; turtles on raw lean meat. Some of the smaller and medium-sized snakes will take earthworms; the medium-sized ones, small frogs and fish; but the large specimens often require live small mammals. For short periods of a few days to a week, most species can fast and remain in good health. For care over longer periods, reprints of an excellent article by Gorham (1963) on care of small herptiles in inexpensive home-made quarters are available through the Education Section of the National Museum of Canada, or directly from the Curator of Herpetology. Other informative publications are those of Breen (1949) on reptiles in general, and Roberts (1960) on turtles; these may be obtained through local pet shops. Some of the handbooks in the section on selected references also contain information on keeping these animals.

For keeping reptiles and amphibians for periods when immediate preservation is not possible and behaviour observations are not required, refrigeration, placing the animals into induced hibernation, is a solution. The best temperature is a few degrees above freezing—about 35°F.

Tadpoles and Salamander Larvae

Tadpoles and salamander larvae may be reared in an aquarium or in shallow pans. Pond water should be used at the start, but evaporation losses may be replaced, as necessary, with tap water which has been allowed to stand a few days in an open container. Aquatic plants may be placed in the container with tadpoles and larvae but are not essential. Tadpoles will feed on algae, aquatic plants, and strained baby spinach. Salamander larvae will eat scraps of raw lean beef, chopped earthworms, or white worms. When the tadpoles and larvae start to transform, a stone or board must be placed in their container to allow them to crawl out of the water. If this is not provided they will drown.

Amphibian Eggs

Amphibian eggs may be hatched in a container of pond water. The resulting tadpoles or larvae should usually be separated into several containers, or a portion of the hatch returned to the pond, to prevent death from over-crowding.

Reptile Eggs

Reptile eggs must have a warm and relatively moist environment and are subject to moulds. Legler (1956) recommends the following method of caring for them:

> "Eggs collected in the field are brought to the laboratory in damp cloth sacks .... Each clutch is identified by a waterproof label and each egg is numbered with a soft pencil. The eggs are then placed between two layers of moist, absorbent cotton in glass containers. The cotton is molded with the hands to form pads approximately one-half inch thick which fit snugly into the container. A finger bowl four inches in diameter at the top and one and three-quarter inches deep was [used]. . . . Molds developed less frequently when a shallow container was used.
>
> "The top layer of moist cotton is tamped down around the eggs and the dishes are stored. A maximum-minimum thermometer is placed nearby to record temperatures during incubation. Moisture can be controlled by sprinkling water over the cotton."

Zweifel (1961) has discussed the difficulties with various techniques and suggested this alternative method:

> ". . . the container is a flexible, transparent plastic bag ($1\frac{1}{2}$ mil polyethylene) and the eggs rest on the medium rather than being buried in it. The exact nature of the incubating medium seems to be relatively unimportant; it acts merely as a moisture reservoir. I have used rotten wood in which skink eggs were found, sand, sandy soil

and sphagnum moss, and suspect that a sponge would serve adequately. The medium is prepared by soaking it in water and then squeezing out most of the water. It is then placed in the bag and the eggs shallowly imbedded in it. Too wet a medium is to be avoided, though the danger is less acute here than if the eggs were buried in the material. The bag is inflated slightly and then sealed with a rubber band.

"Saturation of the atmosphere within the bag is indicated by condensation on its inner surface. Unless the incubation period is very prolonged, it is usually unnecessary to add more water. The general well-being of the eggs can be observed through the plastic, so unless measurements of the eggs need to be taken the bag can be left closed until the eggs hatch."

As young snakes may be able to slit the thin plastic of the bag with their eye teeth, it is advised to keep bags containing snake eggs in a snake-light container.

SHIPPING

Any collections which add to our knowledge of Canadian herpetology may be donated to the National Museum of Canada collection. In addition, any specimens or collections may be sent to the curator for identification. In return, the museum will usually want to keep a portion of such collection, but details may be arranged with the curator by correspondence before the specimens are forwarded. All specimens should be labelled with locality, date, and collector, and a letter giving further details and the collector's address should be sent in the same mail.

Live specimens should *never* be sent without prior correspondence with the curator.

Specimens preserved in formalin or alcohol can be shipped in glass jars, well packed in a cardboard carton or wooden crate lined with excelsior or newspaper. However, a better method is to wrap each collection, with its label, in several layers of cheesecloth or absorbent cotton. The wrapped specimens should then be placed in a plastic bag, and formalin (or alcohol) poured over them. The excess preservative should be drained off and the opening of the bag knotted or securely tied with string or rubber bands. The bag(s) may then be placed in a cardboard carton, lined as stated above. The latter method has the advantage of reduced weight.

All herpetological collections should be addressed as follows:

Curator of Herpetology,
National Museum of Canada,
Ottawa, Ontario.

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CONSERVATION

There is growing concern throughout the world that the accelerating demands of the exploding human population for space and resources will exterminate a large proportion of the once common natural forms of life. There are some who feel that these forms are worth preserving because they have not yet been studied fully and because future generations may deeply regret the loss of such diversity of life.

Reptiles and amphibians have long engendered fear and repulsion in the minds of many people. Because of this they are often killed on sight. Our large harmless snakes, in particular, have suffered a decrease in numbers through this attitude. In many areas, however, they have survived because enough people felt either that their rodent killing abilities justified their existence or simply that since they were harmless there seemed no point in exterminating them for purely emotional reasons. Some have gone further philosophically, realizing that each species has its unique and interesting adaptations for survival which in themselves justify its existence.

However, at this period in history a greater danger threatens species survival than the loss of individuals. The habitats to which these animals are adapted are being destroyed. Swamps and marshes are being drained, forests cleared, and rivers polluted. This is not a new phenomenon—it has occurred on various scales throughout the history of man. However, man is now utilizing more and more of the land, and the scale on which the destruction of the original landscape is being carried on is increasing. Unless some areas are set aside in each region and vigorously protected against exploitation, many species will vanish.

When collecting specimens, it is important to realize that both numbers and suitable habitat are being constantly reduced. If the habitat is still plentiful, a sufficient number of specimens—even several hundred of abundant species—may be collected, if needed, without endangering the survival of a species. However, such numbers of specimens should only be collected when care can be taken in preserving them and when they are to be placed in a permanent study collection. Such collections may even be necessary in order to understand enough about a given species to provide a scientific basis for measures to conserve it, and they do not conflict with the aims of conservation. Obviously, only small collections should be taken of a rare species in a restricted habitat, and no specimens should be collected unless such a collection serves a useful purpose. As noted under the section on cover, the collector should try, as far as possible, to leave his collecting sites in much the same condition as he found them.

Specimens should generally *not* be collected in one area and released in another. Such irresponsible introductions rarely survive, but when they do, introduced individuals may contribute characters from another population to the one into which they are released and may obscure a significantly unique feature of that population. If an introduced species survives, it may obscure the zoogeography of a whole region. If introductions, accidental or intentional, are made, a museum should be informed so that there will be a permanent record of them.

In Canada, the *Canadian Reptile and Amphibian Conservation Society* is concerned with all aspects of conservation and humane treatment of reptiles and amphibians and in promoting a better understanding of their unique place in the natural world. Anyone interested in more information and in supporting the society and its aims should write to Miss Barbara Froom, 8 Preston Place, Toronto, Ontario, or Mr. Alex Findlay, 32 Hartsdale Drive, Weston, Ontario.

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CHAPTER VI

COLLECTING AND PRESERVING OF FISHES

D. E. McAllister

"In collecting fishes three things are vitally necessary—a keen eye, some skill in adapting means to ends, and some willingness to take pains in the preservation of material. . . ." David Starr Jordan
Guide to the Study of Fishes

SUMMARY

1. Drop live into 10% formalin (1 part concentrated formalin and 9 parts water) and slit right side body cavity of specimens over 6 inches long.
2. Do not crowd or bend. A container should be no more than half full of fish and filled to the top with preservative.
3. Include label *in* each bottle of fishes bearing at least: date, locality (body of water, distance from nearest town, county, province), and collector's name; label to be of water-resistant paper written in India ink or pencil.

COLLECTING

Where to Collect

Few places, if any, in the world have been completely collected. Faunas change with climate. As ichthyology reaches higher levels, more refined information is required. There is little danger in overcollecting, particularly when collecting is selective. On the other hand there are many poorly collected habitats and regions which would repay better collecting effort. Examples of this are the larger rivers, the deeper waters of lakes and seas, the northern waters of Canada, and so on. Museum curators can inform one of geographic areas that need attention.

In collecting it is desirable to cover as many types of habitat as possible. Each habitat has its own type of fish life. In a stream, for example, tiny headwaters, broad lower reaches, rapids, pools, backwaters, oxbows, overflow pools, and weed, rock, gravel, sand, and mud sections should all be collected. Springs (hot and cold), caves, and wells deserve attention. Marine habitats worthy of examination include the upper and lower tide pools; rocky, gravel, sand, and mud shores and bottoms, exposed and protected shores, regions of high and low salinity, coral reefs, fishing banks and guyots, surf zones; amongst eelgrass, mangroves, rockweed, kelp, and pilings; surface, mid-depths, and bottoms in shallow and deep waters. Collections from different habitats should be kept separately, and notes should be made of particular niches of each habitat from which fishes came.

Night collecting is well worthwhile as many species remain hidden during the day or remain in deeper waters. There are few night and few winter collections of fishes. A series of collections taken at different times of the year at one station enables studies to be made of movement, growth, and maturation.

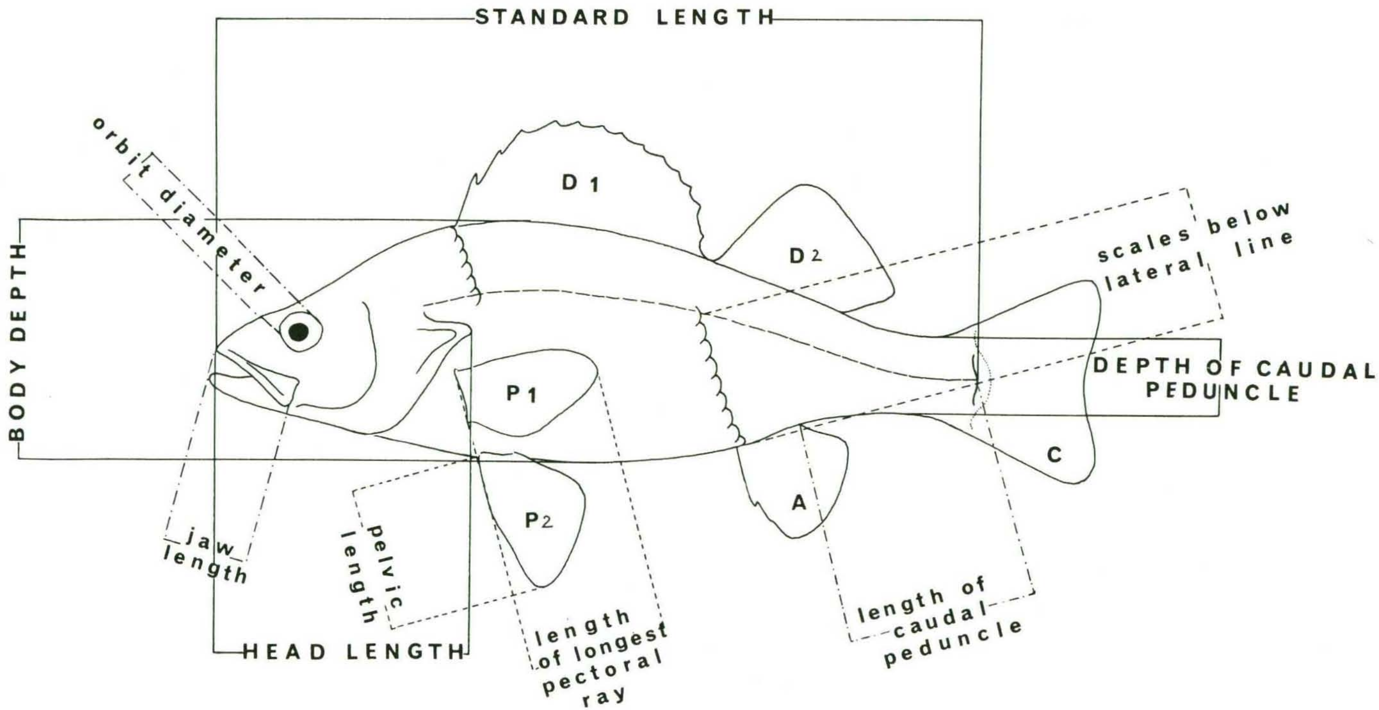

Figure 46. Methods of measuring fishes.

What To Preserve

Specimens of all species at each collecting locality should be preserved. It is wise, as many collectors have learned to their sorrow, to collect a species the first time it is seen; it may not be seen again.

A series of from 30 to 500 of each species from each station is desirable. This number may be modified if it is certain that there are already many of that species from that locale. The large size of specimens or the small quantity of preservative or containers may, of course, impose a limit. Quality, on the other hand, should not be sacrificed to the god of quantity. Better a few fine specimens than numerous, unmeasurable, twisted, scaleless specimens. But better still to take what appears to be an excess of containers and preservatives, particularly when traversing little-known regions.

Specimens of both sexes and of the different sizes available should be collected.

In the case of very large specimens which may not be preserved or frozen whole, the head and the tail and a scale sample may be taken. Photos, scale and fin ray counts, colour notes, measurements of head, jaw length, eye, fin, standard length (snout to base of tail) and total length and weight should be recorded. (*See* Figure 46 for methods of taking measurements.) In the case of sharks upper and lower teeth (or jaws with teeth) and a piece of skin should also be taken.

In the case of rare or endangered species the collector should exercise discretion in the interests of conservation.

Photographs and Colour Sketches

Colour photographs and sketches are a valuable adjunct to collections. Colour in specimens is lost with time, yet it is often a valuable systematic character. Normally the left side is photographed. The fins are spread out (pinned if necessary), the mouth closed. Value is increased by including a photographic colour scale (obtainable at photo stores) and a rule; for convenience the two can be taped together. The collecting label should be included in the photo to record where and when the fish was collected. To obtain sharp focus for scale and fin ray detail, a tripod is necessary. Even lighting without obscuring shadows and a contrasting background gives superior results. A little extra care will result in photos not only of value as a record but of publishable quality. The opportunity may not present itself again to take a photo of a fresh specimen.

Photographing fish in an aquarium enables recording life colour. The fish may be crowded against the front of the aquarium with a plate of glass. This keeps them in one plane for overall focus and usually causes them to extend their fins. In some fishes other views than lateral may be required, e.g., dorsal for rays, batfishes; or several views may be taken with detail shots of interesting structures. Do not overlook habitat photographs.

Good photos may be donated to the National Nature Photograph Collection. The photos will be used for science and education and kept for posterity.

Collecting Equipment and Methods

Each new collecting method brings in new species. Only certain methods will work in certain habitats. A variety of collecting methods is therefore rewarding. Several reviews of fishing gear are worth consulting for ideas: Umali (1950), Okada (1959), and Bean (1887).

Nets

The strength, durability, resistance to rot, and invisibility of nylon nets make them superior to those of cotton or linen, although they are more expensive. Braided lead and float lines where leads and floats are enclosed are much less likely to tangle.

One of the most useful of collecting tools is the *seine*, a net of fine mesh pulled along the bottom. It is weighted to sink when in water deeper than its depth. Those of smaller meshes catch smaller fish but are harder to pull. Those with a built-in bag in the middle are the most productive. Generally a mesh of one-eighth to one-quarter inch is most effective. One with a fine-meshed bag and the rest of the net of coarser mesh combines these two qualities effectively.

Two seines may be found more useful than one. A small seine, about 12 by 6 feet, may be used for small streams and embayments. A 40 or 60 by 6

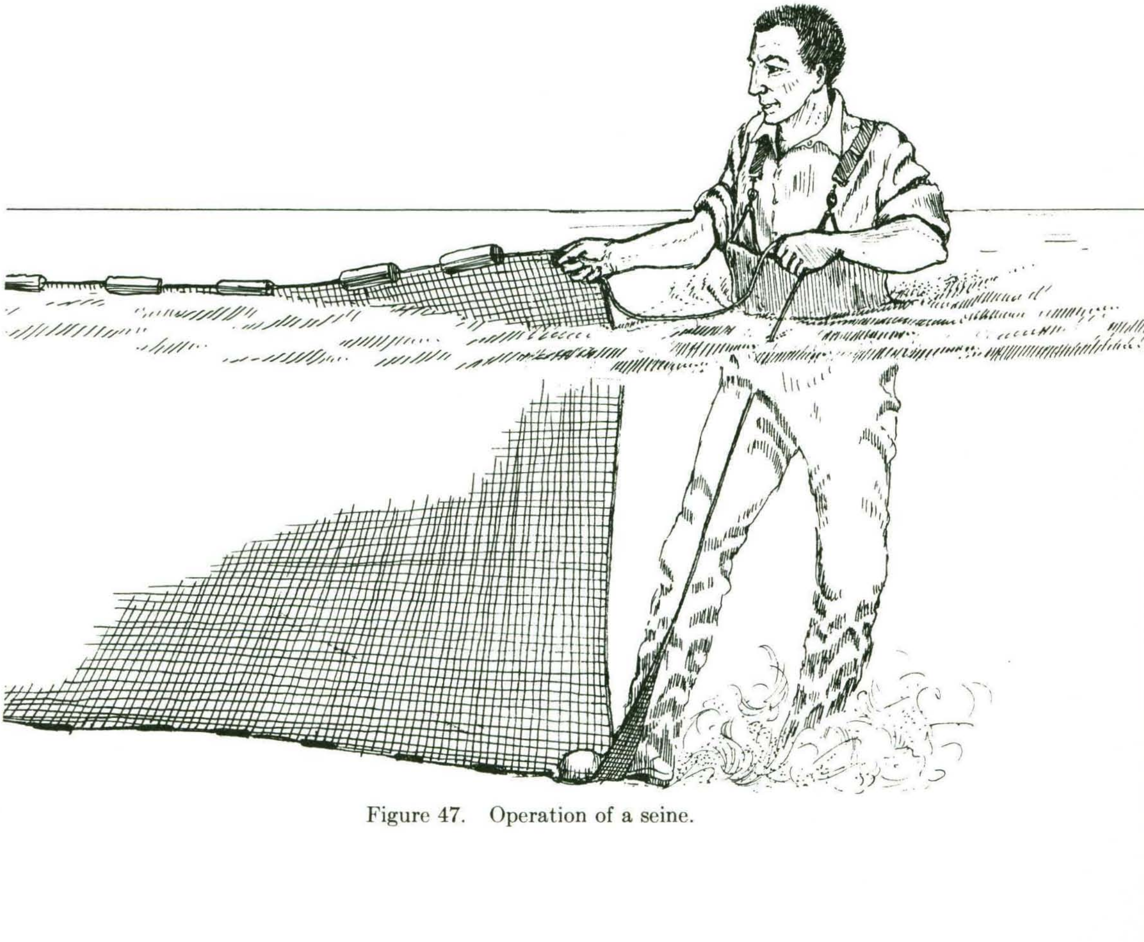

Figure 47. Operation of a seine.

or 10 foot seine with a bag may be used for broader beaches of lakes, rivers, and the sea. These nets require two persons to pull them. The bottom of the loop of rope joining the top and bottom of the net may be placed underfoot (Figure 47) to ensure that the lead line stays on the bottom. Force to move the net is applied by leaning forward and pulling with the arms and shoulders. Alternatively the seine may be operated with poles tied to the ends. The net billows out behind, forming a bag. Seining downstream, except in fast water, is said to be more productive. In riffles and rapids the net may be fastened across the current and the fish driven down into it by stamping and by kicking boulders and other cover. Creosote applied upstream may drive the fish down into the fixed net.

Seines larger than 60 feet may be used but may require more men or a winch (unless seine of quite large mesh). Large seines may be set out in deep water with a boat and then pulled into shore with long ropes, thus seining a large undisturbed area. When pulling seines through vegetation, care should be taken to avoid rolling up the bottom of the net.

Purse seines are large seines which are set out in a circle on the surface. The bottom of the net is then closed or pursed by tightening a line which runs through rings at the bottom of the net, thus trapping fish within the circle. By drawing the net into a boat the fish are gradually restricted into a small pocket from which they can be dipnetted.

Dip nets are often handy to catch fishes attracted to a *night light* or bait, in spawning swarms or those killed by explosives or ichthycides. A metal hoop with the netting laced inside with wire makes for durability.

Gill nets, open-meshed nets which catch by the gill covers or other projecting body parts, may yield many specimens. Best results are obtained by setting overnight and retrieving early in the morning, the meshes being less visible to the fish at night. Frequent removal of fish from the net means that specimens are fresher, less abraided by the meshes, less eaten by isopods or other scavengers or torn by predators. The nets may be set floating at the surface, hung in midwater from the surface, or set on the bottom. To make nets easily retrievable buoys should be used; by so marking the position of the nets, boats can easily avoid disturbing them. Setting nets out at right angles from shore is often productive.

It is best to join several nets of different mesh sizes as gill nets are selective in the size of fishes caught, e.g., $\frac{3}{4}$-, 2-, and $3\frac{1}{2}$-inch (stretched mesh) panels may be joined. The length and depth of the net may be varied according to requirements. However, if a standard gang of nets is set in a standard fashion, catches from different places may be compared.

McKenzie (1947) and Sprules (1949) described the *Canadian prairie ice jigger* used for setting gill nets under the surface of the ice. By using it, only a single pair of holes need be made in the ice. Pulling on a line causes the jigger to work along under the ice. When it has gone far enough, a second hole is made over the jigger. The net can then be pulled along until set between the two holes by means of the rope threaded by the jigger.

Trawl nets or trawls are bag-like nets which are generally drawn through the water by a boat. They provide a most effective collecting method on smooth bottoms or in mid-water, even in the deep sea. The bag may be kept open by planing boards—otter trawls; by a pole—beam trawl; or by vanes—mid-water trawls. Long trawl drags should be avoided as they may damage specimens. Trawls are themselves damaged on rough bottoms and work best on gravel, sand, or mud bottoms. An echo sounder is a valuable aid in trawling. Isaac and Kidd (1953) and Barraclough and Johnson (1956, 1960) report on mid-water trawl design. Baldwin (1961) presents a design for a trawl to use from small boats.

Plankton nets are useful for catching larval fishes. The one metre stramin net will catch the adults of small species. *Throw nets* are valuable in certain situations.

Hook and Line

The ancient art of fishing with hook and line may be employed with profit. Angling with various lures or bait from boat, shore, or through ice is too well known to require description. Baited set lines may be floated at the surface, at mid-depths, or on the bottom.

Explosives

Under certain conditions, explosives, such as dynamite, may prove suitable. In using dynamite the cap and fuse should be sealed into the stick with heavy grease. Hubbs and Rechnitzer (1952) and Ferguson (1962) discuss effects of explosives on fish life. Schwartz (1961) provides a bibliography on the subject. In general, explosives kill only fishes with gas bladders or fishes close to the explosion. As only some of the killed fishes float, it is necessary to make arrangements to collect those specimens that sink to the bottom. Naturally due precautions should be taken, legal requirements should be followed, and public relations should be considered in the use of explosives.

Ichthycides

Numerous substances toxic to fishes may be employed in collecting. Rotenone, harmless to man or livestock, under proper concentrations is perhaps the best. It is prepared from derris, cube, and timbo root (chemical formula $C_{23}H_{22}O_6$) together with a dispersing agent. In using the powdered form it is generally mixed with water into a liquid or paste before dispersal. The emulsified form is ready to use. The concentration employed depends on the temperature, water quality, and so on. It may be from .5 to 1 part per million by weight, but the instructions supplied should be followed. In streams it is applied for a period of about 15 minutes so that fish downstream are exposed continuously. A net is stretched across the river downstream. It should be applied just above a point where it will be diluted below toxic level by the confluence of a tributary. Rotenone, unlike the more dangerous toxaphene, breaks down fairly readily. Quantities should be carefully calculated on volume flow.

Ichthycides may also be employed on lake or sea shores; in small embayments; in reed patches, tide pools, coral reefs; underwater with plastic bags or squeeze bottles; and even in surf.

Other ichthycides—copper sulphate, lime, formalin, and so on—may be employed in restricted situations such as pools. Many native peoples know of local plants that will drug or kill fishes.

In all ichthycide operations it must be remembered that water is a common resource of all. It should not be applied in sport fishing waters, nor in drinking or swimming water, and never in waters with rare or endemic organisms.

Scuba

Scuba gear is of great value in collecting and observing fish. With foam rubber suits, one may enter even Arctic or Antarctic waters. Useful underwater equipment includes spears (hand or powered with rubber), springs or compressed gas, ichthycides, explosives, underwater cameras, traps, depth metres, and so on.

Spears are, of course, of value above water, particularly in association with spawning runs, night lights, and certain large fishes.

Electric Fishing

Large and small portable back-pack electric shockers are of considerable value in collecting. AC or DC units may be employed. Their value lies in the fine specimens collected, in their usefulness on snag or bouldery bottoms, and in the selective nature of catching. (*See* Burnett (1952, 1959) or bibliography in Schwartz (1961).)

Traps

Traps vary in size from small minnow traps to huge pound nets and weirs. The fish may enter traps out of curiosity, be attracted by bait or electricity, or impelled by stream or tidal current. Most traps employ a funnel entrance which guides the fish in, and from which the fish find it difficult to escape.

Local fishermen and small boys are often of great assistance to the collector. The game warden and experienced local angler should not be overlooked, nor should the commercial fisherman.

Check List of Collecting Equipment

The following list summarizes equipment necessary on an ichthyological expedition:

Nets: seine, gill, dip, trawl, plankton
Hooks and line, lures, bait, sinkers, gaff
Explosives
Ichthycide
Scuba equipment: spears, depth gauge, watch, mask, fins, suit, weights, tanks
Fish shockers
Traps, dredges
Rope, cord
Weighing scales, tape measure
Echo sounder, thermometer
Salinity gear, water quality kits
Camera, film, close up lens, tripod
Aquaria, oxygen tablets
Waders, bathing shorts, weather clothes
Cheesecloth, hypodermic syringe and needles
Formalin, alcohol, alcoholometer
Graduated cylinder
Labels, notebooks, data sheets
Stationery, collecting permit
India ink, pens, pencils
Identification books
Maps, charts, pilot book, navigation gear
Boats, oars, anchor, life preservers
Engines, spare parts, fuel, lubricants, shear pins
Floodlight, gas lanterns, flashlights
Food, drink, matches, portable stove, utensils
Camping gear, tent, sleeping bag
Mosquito lotion
Sunglasses, first-aid kit and book
Ice drill, ice chisel, ice jigger
Bottles, cans, drums, plastic bags
Knives, scissors, scalpels, saw
Soldering iron, hammer, nails, screw driver

PRESERVING

Containers

Containers for fish specimens should ideally be unaffected by water, formalin, and alcohol; they should seal tightly but open easily, and each should have a mouth as wide as the container itself to admit broad fishes. Iron, brass, and copper containers are *not* suitable. Glass bottles and polyethylene bags, jars, and refuse cans are excellent. Plastic tops are preferable to metal ones. For short periods of time lacquered or tinned cans and drums may be used. Ordinary cans and drums may be used by lining them with plastic bags. Five-gallon cans with wide lids are a convenient size. Fibre-glass containers when made with an alcohol-proof base are excellent. New barrels of light-coloured wood may be used for temporary storage, except in hot countries.

Shipping

Only after fixing about a week, may specimens be shipped. Collections of small fishes may be wrapped with a label in gauze and tied lightly with string. Burlap or other coarse cloth is not suitable. Larger fishes should be wrapped with a label in gauze, individually. Or plastic bags with a small amount of fluid put in each bag may be substituted for gauze. These parcels are then lightly packed in a waterproof container with a small amount of fluid. Empty space is filled so that specimens do not knock about. Fill with cloth, white paper, or white wood chips. The container should be sealed. If shipped for long distances metal containers should be soldered closed as a surprising amount of fluid may evaporate through a small hole.

Small lots can be conveniently mailed in cans sealed with a home-canning machine, in sealed plastic bags (except for spiny specimens), inside boxes or mailing cartons.

An address label should be included on the inside and the outside of each shipment. An outer label should also state "Scientific specimens of fishes, no commercial value," with the addition of "Replacement value $----" in the case of rare or valuable specimens.

Preservatives

Two standard field preservatives are used, 10 per cent formalin and 70 per cent ethyl alcohol. Alcohol is bulkier to take into the field and preserving in it requires more time and care; however it is more agreeable to work with and will not freeze in cold weather. Alcohol is also more expensive (unless the tax is rebated to the institution), and there may be legal problems. Formalin is less bulky to carry into the field, requires less trouble in preservation, gives greater certainty of preservation; but it is less agreeable to work with and freezes in cold weather. Most modern collectors use formalin.

In the museum, specimens are stored permanently in 45 per cent isopropyl alcohol or in 70 per cent ethyl alcohol (occasionally in 10 per cent *buffered* formalin). If the specimens have been in formalin, they are soaked 24 hours or more in water to wash it out before they are transferred to alcohol. Colours keep better when specimens are stored in the dark.

Formalin (Formol)

Commercial concentrated formalin usually consists of 37 per cent formaldehyde gas (CH_2O) in water plus some methyl alcohol to prevent polymerization. It is poisonous, irritating to inhale, and painful to abraided skin. A 10 per cent solution (*of the concentrated form* by volume) is made by adding 1 part of concentrated formalin to 9 parts of water.

As formalin is acidic and decalcifies bones and scales, it should be buffered particularly when specimens are left in it for some time. This may be done (Miller, 1952) by adding:

(a) to $\frac{1}{2}$ gallon of 10% formalin 1 level teaspoonful of borax *or*

(b) to 1 pint of concentrated formalin add 2 ounces of hexamine and 6 pints of water *or*

(c) to 1 pint of concentrated formalin add 5 pints of water and 3 ounces of concentrated ammonia solution.

If it is necessary to closely examine specimens from formalin, they may be deformalized by a few minutes immersion in a solution made by addition of 1,260 gm of $NaHSO_3$ and 840 gm of Na_2SO_3 to 4.5 gallons of water.

Paraformaldehyde

The white powder paraformaldehyde is a polymer of formaldehyde. It can be converted to formalin. Because of its small bulk it is convenient to take on long expeditions or air trips, or as a source of emergency preservative. To prepare 10 per cent formalin from paraformaldehyde, 16 grams of paraformaldehyde and 4 grams of anhydrous sodium carbonate are mixed. A small amount of wetting agent, such as Alconox, aids in dissolving these. It may be packed in vials or polyvinyl film (such as Saran-wrap or Parafilm) packs. Each packet mixed with 400 ml of cold water will produce a 10 per cent buffered formalin solution (slightly alkaline but good for temporary field storage). When mixed with hard water, a slight but harmless precipitate of calcium carbonate may be formed.

Persons sensitive to formalin are cautioned against the toxicity of paraformaldehyde, especially against inhaling its dust. (This section on paraformaldehyde paraphrased from Huheey, 1963, and Taub, 1962.)

Ethyl Alcohol

Ethyl alcohol is also called ethanol, grain alcohol, or spirits of wine. (Wood alcohol, methyl alcohol, or methanol as it is variously called is a poor preservative; it is poisonous, while undenatured ethyl alcohol is not.) Ethyl alcohol is ordinarily obtained as 95 per cent (66.6° overproof) unmatured undenatured ethyl alcohol. To preserve fishes in ethyl alcohol, the preservative must be gradually brought up to the 70 per cent concentration; otherwise the fishes will shrink. An alcohol hydrometer or alcoholometer is essential for preserving with ethyl alcohol. The alcohol percentages given below are by weight, but for simplicity the volume measures necessary to make them up are given. The following concentrations are required for preserving:

(a) 50% (=proof spirit) made with 9 volumes of water to 10 volumes of 95% alcohol

(b) 60% alcohol (20° overproof) made with 6 volumes of water to 10 of 95% alcohol

(c) 70% alcohol (40° overproof) made with 3 volumes of water to 10 of 95% alcohol.

(*See also* the British Museum Instructions for Collectors No. 13. Alcohol and alcoholometers.) Note that when alcohol and water are mixed, some shrinkage in total volume occurs.

Freezing

Quick freezing (best at about 10° F) is an effective mode of preservation. Colour is retained. Large specimens can be dealt with by freezing. The specimens may be thawed out later in 10 per cent formalin, or in water and then fixed in formalin.

Drying. (*See* Other Preservatives.)

Other Preservatives

For histological work, special preservatives such as Bouin's may be required. In the case of emergencies, the following preservatives may be resorted to:

(a) strong alcoholic drinks such as whiskey, rum (best, light not dark), gin, arrack, or alcohol-based shaving lotion. Sake, wine, and beer are not sufficiently alcoholic.

(b) strong brine solutions—one pound of salt to one gallon of water.

(c) skinning and drying the skin, particularly in the case of very large specimens. Because of its extreme toxicity the use of arsenic is *not* recommended as a preservative.

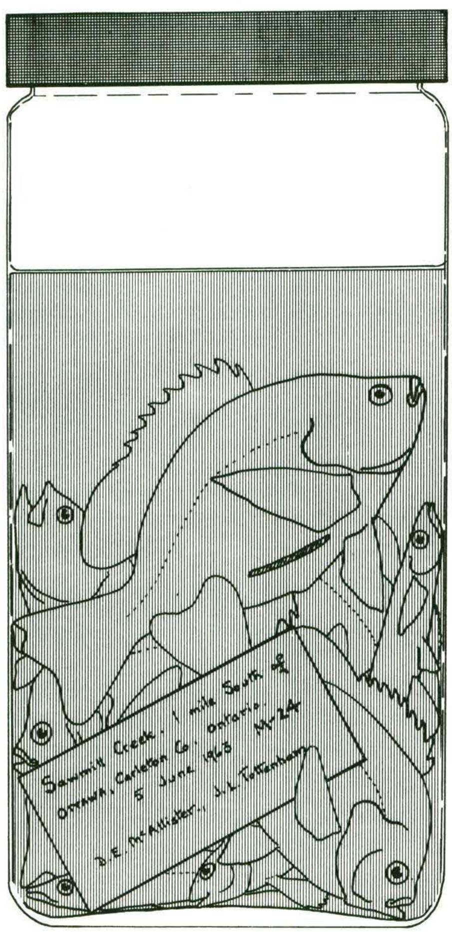

Figure 48. A preserved collection of fishes.

Preserving

In preserving specimens, those from a single collection (one place at one time seine-haul or net set) are usually preserved and shipped together to the museum. Sorting to species is done later at the museum.

In preserving specimens the following points should be observed. Specimens should not be bent or crowded. While preserving, not more than half the container should be filled with fish. An effort should be made to have the fins extended and the mouth and gill covers in their normal position. A slit made into the right side of the body cavity will ensure preservation of internal organs (*see* Figure 48). Avoid cutting internal organs. Very large specimens may require injections of preservative into the muscles of the back; or better, slits into the back muscles from inside the body cavity.

Delicate larval or oceanic fishes, or those with deciduous scales may be lightly wrapped in gauze or kept separately in vials. Larval and small oceanic fishes may be preserved in more dilute formalin—5 per cent. Fishes with large spines or rough scales should be separated from the rest of the collection.

Under certain conditions the strength of the preservative must be increased, e.g., tropical climate; specimens beginning to decay; very large specimens; and containers completely filled with fish (instead of half-filled).

With a bit of extra care in handling, very fine specimens may be obtained. These will be easy to identify, easy to obtain data from, and may last over a century. It is therefore well worthwhile. If specimens are laid out flat in a pan, mucous, blood, sand, and vegetation may be brushed off. The specimens may be laid straight and the fins spread, and then covered with formalin. While fixing them in the pan, colour notes may be made. When the specimens are firm, they may then be carefully placed in jars filled with fresh preservative.

Filling containers to the top lessens slop of fluid and wear and tear on the specimens during transport. It also adds extra protection against evaporation.

Formalin

The normal procedure in formalin preservation is to carry to the collecting site a number of bottles or cans suitable in size, partly full of concentrated formalin. The live fish are dropped into the concentrated formalin. This kills them quickly and causes the fins to spread. The freshwater or seawater is added till the bottle is full (then the label).

Ethyl alcohol

When preserving with ethyl alcohol the following steps are taken:

Fish are put (a) in 50% alcohol for a few hours to 2 days (the shorter period in high temperatures);

(b) then in 60–70% alcohol for 7 to 14 days depending on specimen size. (Alcohol should be tested at least every other day and fluid changed or brought up to strength);

(c) then in 70–75% alcohol for final storage. (These steps on preserving in alcohol from the British Museum Instructions for Collectors.)

Ethyl alcohol can be reused if filtered and brought up to strength, or if redistilled.

Drying Skins

This method is included for interest's sake or in case other methods are not possible. Fish specimens have, in the past, been dried on herbarium-type sheets. The following steps should be taken:

Fish should be (a) cut in half, removing gills, brain, and intestines but leaving median fins with the left half intact;

(b) washed and gently rubbed dry;

(c) placed on board, with cut face downwards, pin fins expanded;

(d) exposed in sun or before fire, then flesh side exposed till dry;

(e) placed between papers and pressed flat, after skin has been readily separated from flesh;

(f) laid on parchment (or perhaps plastic sheet), scales down to prevent sticking to exuded glutinous matter. The sheet should be replaced in 1–2 hours. Then skin with fins should be mounted on sheet of paper or cardboard and varnished, if desired. The process takes about 24 hours. A similar method has been used in herpetology to preserve skins for study of colour pattern. The description of this method has been taken from Wheeler (1958). (*See also* Bertin (1954).)

Sodium Ascorbate

A paper by Yoshida(1962, Bull. Misaki Marine Biol. Institute, Kyoto U. (3): 67–68, 1 col. pl.) was received in the final stages of the present paper. It describes the following method of retaining the colours red, orange, yellow, brown, and black in preserved specimens.

The solution below is injected into the body after the gases in the visceral cavity are exhausted. Fish immersed in the solution, put in a vessel kept tightly covered with a lid sealed with electron wax, after the vessel has been exhausted of air in it.

5–10 ml conc. formalin
0.3–0.6 g sodium ascorbate
100 ml of 1% NaCl solution or 30% seawater.

Mounting and Casting Specimens

Since mounting and casting specimens is more in the realm of display than science, this aspect will only be briefly dealt with. A mould may be made of plaster of Paris using the specimen. From this mould a hollow cast is made. To it are attached the original or artificial fins of plastic. Oil paints with an iridescent pearly essence lacquer are used to recreate the original colour. The final cast may also be made of celluloid or liquid rubber. Alternatively the fish may be skinned and stuffed, and the colours may be restored. (For details *see* Mackay (1958), Migdalski (1960), or Royal Ontario Museum (1958).)

LABELS AND RECORDS

The labels and records on collections are almost as valuable as the specimens themselves. It may seem obvious that labels and records should be written on proper paper and in legible writing at the time of collection. However many collections are received with labels half dissolved, or with "arr is n L. near erndl . May 19 ," or labelled "Trout Lake" (there being about fifty Trout lakes in Canada!). From adequate field notes another person can collect at the identical spot. Notes provide knowledge on the ecology, life history, and colour of the fish.

Labels

Labels should be included *in* the container with the specimens. The label will then less likely be lost, obscured, or torn, as happens to outside labels. Since the label is in the fluid, it must be written on stout paper which will not disintegrate.

The paper must accept India ink and pencil. A label should have at least exact locality, date of collection, number of collection, and collector's name. Additional data are written on record sheets or in a field notebook under the same collection number. When rough-scaled fishes are included

NATIONAL MUSEUM OF CANADA
DIVISION OF FISHES
CATALOGUE SHEET

Province or Country Quebec Coll. No. 0-28
County: Drainage
Locality: Lac Deschenes, Ottawa River, 1½ miles southwest of Champlain Bridge Hull
Lat. 45° 27' N., Long. 75° 44' W.
Water: clear, unpolluted Salinity: — ‰
Vegetation: lily pads and reeds
Waves: calm
Bottom: mud and boulders
Cover: weeds and boulders Temp. 23° shore, 17.5 at 50' out Air: —
Shore: grass flat Current: none
Dist. offshore: 0-50 feet Stream width: 1 mile
Depth of capture: 0-3 feet Depth of water: 0-3 feet
Collected by B. A. McKenzie, F. R. Cohen, J. L. O'toole
Tide: — Date: 17 Sept. 1963
Method of capture: 30 x 6 foot x 1/8 inch mesh seine with bag, 2 hauls
Orig. preserv.: Formalin: 10% Time: 1300-1400 hours
all specimens kept. Sunny day: 2 jars.

<u>Ictalurus punctatus</u> (1)
from fisherman, caught on spoon about 200 feet off shore
<u>Fundulus diaphanus</u> (3)
in water 1-3 inches deep next to shore where 22-23°C.

<u>Ambloplites rupestris</u> (6)
with red eyes
<u>Lepomis gibbosus</u> (5)
with red spot on operculum

<u>Etheostoma nigrum</u> subsp. (1)
noticed in bouldry section.

<u>Micropterus</u> seen but not caught.

– Habitat photo taken –
Catfish preserved in separate jar

Figure 49. Example of a data sheet, freshwater specimens.

in the collection, the writing may rub off. In such collections the label may be placed under the gill cover, in the mouth, or in a small plastic bag or vial. If tags are tied onto specimens it must be ensured that the label is stout enough not to tear off.

Labels may be made of the following: a high-quality, high-fibre-content paper; a really good quality pure rag paper; linen ledger paper; best quality of bond letter paper. Suitable paper or label cards may be obtained from paper suppliers or from the museum. Paper should be specified as alcohol- and water-proof, able to accept pencil and India ink. *Test* before using.

Labels are best written in India ink, although pencil would do. The India ink must be alcohol-, formalin-, and water-proof. Since the ink must be dry before it is placed in the preserving fluid, a rapid drying ink is desirable. The following brands have been found suitable: Chin-Chin (best), Talens Rembrandt, Higgens engrossing—all black water-proof type inks. If pencil is used, a medium or soft one is preferable since a hard one makes a fainter impression.

NATIONAL MUSEUM OF CANADA
DIVISION OF FISHES
CATALOGUE SHEET

Province or Country off Newfoundland Coll. No. B M-25
County: – Drainage –
Locality: e Grand Banks about 250 nautical miles
Southeast of Cape Race, southeastern tip of Newfoundland
Lat. 45° 30′ 00″ N., Long. 48° 28′ 00″ W.
Water: – Salinity: 35.69 ‰
Vegetation: none
Waves: calm
Bottom: sand and mud
Cover: none Temp. 8.7° C. Air: 15.4°C.
Shore: – Current: –
Dist. offshore: 250 nautical miles Stream width: –
Depth of capture: bottom Depth of water: 100 fathoms
Collected by B. A. McKenzie on the Fish. Res. Bd. "A.T. Cameron"
Tide: – Date: 17 Sept. 1961
Method of capture: 20 foot otter trawl with 1 inch mesh, ½ inch cod liner
Orig. preserv.: Formalin: 10% Time: on bottom 0955-1025 hours,
½ of specimens kept, representative species and size sample

Raja radiata (15)
2 mature males
Mallotus villosus (20)
males with elongate fins and lateral ridges, backs olive green, sides silvery.

Hippoglossoides platessoides (32)

Triglops murrayi (3)
males with genital papillae and black spot at front of dorsal; both sexes with black spot in rear of dorsal and irregular broad ~~black~~ dark marks on sides of light brown, eye with gold tint.

— Colour photo of each species taken —

Figure 50. Example of a data sheet, marine specimens.

Records

It is easier to use data record sheets, such as the ones in Figures 49 and 50, illustrated below, than to record data in a lined notebook. Using data sheets is quicker, and a standard set of essential data is then recorded for each collection. The data sheets are punched for inclusion in a ring binder, preferably one with a water-proof plastic cover. Abbreviations should be avoided—only *you* may be able to decode them.

The following notes explain details to include under the headings on the data sheets. Not all categories apply to all collections; there are no tides in most lakes. *Province* or *Country*, *County* are self-explanatory. *Coll. No.* is an abbreviation for collection number. *Drainage* refers to the river system, e.g., Ottawa - St. Lawrence. Under *Locality* should be given the body of water (stream, bay, lake), its air distance from the *centre* of a nearby city or town, the highway it is on or near, the township, the regional name, and the exact latitude and longitude. Under *Water* may be described its clarity, hardness, pollution. The *Salinity* of seawater is noted in parts per thousand. Under *Vegetation* is noted the amount and kind of aquatic plant life. Under *Waves* may be noted the degree of exposure or wave height. The type of bottom, bedrock, boulders, sand, gravel, mud, silt, detritus, and so on, is stated under *Bottom*. *Cover* refers to places where fish may hide, such as rock crannies, overhanging banks, sunken logs, coral heads. Under *Temp.* is given the water temperature or the *Air* temperature or both. Under *Shore* is indicated the type of shore: such as shelving sand beach, cliff, tidal flats. Under *Current* is indicated the water velocity of streams or tidal currents, if possible in feet per second; if not, as rapid, medium, slow, still. *Distance offshore* and *Stream width* are self-evident categories. The *Depth of water* is the depth from the surface to the bottom, and the *Depth of capture* indicates where the collection was made, i.e., at the bottom, mid-depths, or surface. Under *Collected by*, the collector(s) are listed with the head of the party first; the organization or the name of the vessel from which the collection was made may also be included. Under *Tide*, the level of the tide at the time of collection should be stated and, if pertinent, the level of the tide pools above, indicated. The *Date* should be written in full, e.g., 11 September 1962, so that there will be no possibility of error. *Method of capture* indicates the collecting method. The size and mesh of net, the length and number of hauls, should be indicated so that an approximation of concentration of fishes may be made. Under *Formalin* may be written 10 per cent, or if other preservatives were used indicate under *Orig. preserv.* Under *Time* is indicated the hours spent in collecting for the purposes of determining daily habits of fishes and for estimating a catch per unit effort, e.g., 1400 to 1445 hours. In benthic trawling, one can state the time the trawl was on bottom. In regard to 'catch per unit effort,' the proportion of the catch retained may be stated on the first blank line. Under particular species, it might also be stated whether 'all kept' or 'half kept.'

The blank lines below allow for flexibility not permitted by the upper half of the sheet. If possible a list of the species with the number of specimens of each is made below. Under each species, note their particular habitat, behaviour, condition, maturity, colour, and so on. The back of the sheet may be used if necessary. If a collection is split and placed in two containers, a note should be made of it on the data sheet. Habitat photographs may be attached to the data sheets, but photos recording colour of fishes should probably be kept in a main photograph file.

On expeditions a running diary should be kept of places visited and the times. Some duplication between the diary and the data sheets enables a check on errors or omissions. Collection sites may also be noted on a map which can be kept with the diary.

Local names of fishes are worth gathering and may be recorded in the diary or in the data sheets.

Some general works on collecting and preserving will serve as additional sources of detail: the British Museum Instructions for Collectors No. 3 and No. 13, the introduction to Hubbs and Lagler (1958), and Smith (1950). These and mimeographed instructions of the Institute of Fisheries, University of British Columbia, Royal Ontario Museum, and Stanford University were of assistance in preparing these instructions.

Licences

In some provinces it is possible to make small collections with a small minnow seine and by angling without licence or only with an angler's licence. For provincial freshwater collecting, the provincial fish and game or resource departments issue scientific collecting permits to responsible individuals. In other provinces or for broader collecting or for marine collecting (except Quebec) scientific collecting permits may be obtained through the Department of Fisheries, Ottawa. In Quebec, freshwater and marine fisheries are under provincial authority. Listed under Game and Fish Regulations in the current issue of the Canadian Almanac and Directory are the names of whom to write to for permits. When collecting in any locality, one should inform the local fisheries official not only out of courtesy and legal requirements but because of valuable information he may be able to give.

DEPOSITION OF SPECIMENS

After studies have been completed, specimens may be deposited in museums. These specimens will then provide a permanent record of the study as a basis for future studies and also enable that study to be extended. Many museums, including the National Museum, appreciate donations of specimens. Canada is an enormous country from which to secure adequate geographic representation. Containers and preservatives may be given by museums to persons collecting in little known areas. Private collections may be willed to the museum for integration with the national collection for the future use of science and education. Holotypes should be deposited in museums having a permanent ichthyological collection and curator, for example, the National Museum of Canada.

Many museums, if requested, will identify or verify identification of fish specimens. It is customary for the museum to retain half the specimens sent for identification, but other arrangements can be made with the curator. Inquiries and donations may be addressed to the Curator of Fishes, Natural History Branch, National Museum of Canada, Ottawa, Canada.

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CHAPTER VII

COLLECTING SKELETONS

PREPARATION OF ROUGH SKELETONS

The collecting of skeletons, apart from the skulls, has been too much neglected by zoological collectors. The bones of an animal are very important to the zoologist, as they show the deeper relationships and mode of life far better than the outward parts. The bones of modern animals are necessary to the palæontologist for comparison with the extinct animals of the past, which are seldom known by more than their bones. The modern taxidermist also needs to study the skeleton in order to mount his specimens properly, as the skeleton shows the possible limits of size and movement.

Preparing skeletons is a simple operation, consisting merely of cutting away the flesh as cleanly as possible after the skin and viscera have been removed. Skeletal material can often be made from a specimen that is in poor plumage or pelage, or has spoiled so that the skin is not suitable for preservation. Saving the bones of such a specimen is a clear gain to science.

In the science of osteology (the study of bones) skeletons are classified as disarticulated or ligamentary. Skeletons that have bones large enough to be drilled, wired, and bolted together in mounting are disarticulated. If the bones are too small or delicate to be treated in this way they are cleaned but left attached together by their ligaments. Birds, snakes, lizards, and mammals up to the size of a fox are generally treated as ligamentary skeletons. The following methods are used by our museum collectors, adapted from the directions of the late Dr. F. A. Lucas, past master of osteological preparation (1891, etc.).

If possible, the sex and the usual scientific measurements of the animal to be treated should be taken. The date and locality should also be recorded. A catalogue number should be given to the specimen, and a numbered tag of metal, leather, wood, or strong manila paper attached to each separate segment into which the skeleton may be divided.

It is ordinarily impracticable, though not impossible, to make a complete skin and skeleton of the same animal. If the species is rare or unknown to the collector, take the skin off as carefully as possible and preserve it for identification. Otherwise, remove the skin roughly and also all the internal soft parts. Do all the work with a knife, as hatchet or saw should never be used on skeletons. Be very careful not to cut the breastbone or any cartilages attached to it, particularly the disk-shaped piece with which it ends.

If the animal is smaller than a fox leave the limbs attached to the body for convenience until the flesh is cut away. The quickest way is to cut the large tendons near the joints and then pull the flesh loose. Be careful not to cut the patella (kneepan) and leave it embedded in the large tendons attached to the tibia or femur.

Be careful not to lose the clavicle (collar bone). In the cat family the collar bone is very small and lies loose in the flesh between the scapula (shoulder blade) and the front end of the sternum (breast bone). The collar bone of a weasel is very minute and difficult to find, whereas that of climbing and burrowing animals is usually well developed, uniting the shoulder blade with the breast bone. Deer, antelope, bears, and seals have no collar bone. If the legs are detached, leave the collar bone attached to the shoulder blade.

Next, disjoint and clean the skull, being careful not to break any of the neck vertebræ in the operation. In removing the eyes be careful not to break the slender zygomatic process (cheek bone) just below the eye and do not punch a hole through the thin bony plates back of the eyeballs.

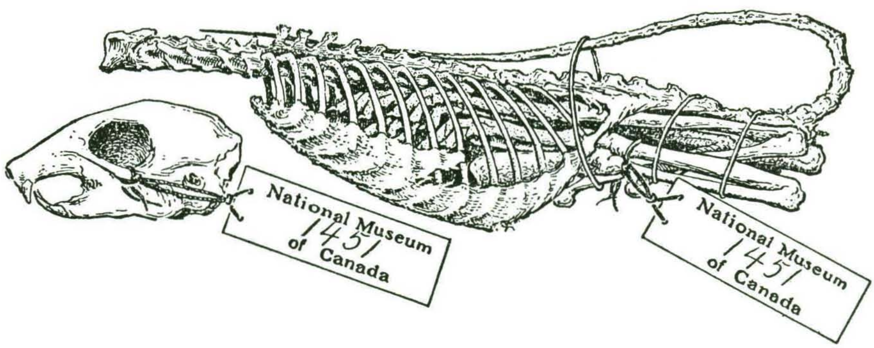

Figure 51. Ligamentary skeleton of small mammal cleaned and dried for packing.

Also, be careful not to break any of the thin bones on the base of the skull back of the upper teeth, particularly in deer and other ruminants, and be careful not to cut or scrape off any bony projections.

Remove as much of the brain as possible with a scraper, bent wire, or small stick. A green switch with the end pounded into a fuzzy brush is useful and loose brains may be washed out with a syringe or by pouring water through the foramen magnum and shaking the skull vigorously.

When cleaning the ribs, take care not to cut the cartilages joining them to the breast bone, and when the tail is reached, look out for a few little bones projecting downwards from the first few vertebræ.

Fold the legs snugly along the body, or, if they have been detached, thrust them, with the skull, as far as possible into the chest cavity, bend the tail back upon itself, and tie the whole into a compact bunch with strong thread or twine wrapping (Figure 51). Any detached bones, or splinters of broken bones, should be rolled up in a rag and tied firmly to any of the leg bones.

Hang the skeleton to dry in the shade, or if necessary in the sun or by a fire, and away from dogs, cats, and rats. If the parcel is to be some time on the road, give it a very thin coat of arsenical soap to repel insects, or, better still, soak the skeleton for an hour or so in the sodium arsenite solution (*See* page 75); all rough skeletons in collections should be thus treated. On short collecting trips the poisoning may be omitted, but if the bones are left for some time uncared for, insect larvæ are apt to eat the ligaments and mix things up generally.

Special Points Regarding Skeletons

The bones of the hyoid apparatus should be carefully saved, but are generally lost by inexperienced collectors while removing the tongue. The hyoid bones are attached to the larynx and connect the windpipe with the base of the skull (Figure 52).

Small bones, called sesamoids, are usually found embedded in the tendons where they ply over the under sides of the toes, and in other places, so the tendons should not be cut off close to the bone. There are often one or two small bones on the back lower part of the femur (thigh bone), and these should be left in place. Rabbits have a slender projection extending backward at the lower end of the shoulder blade, and this should not be broken off.

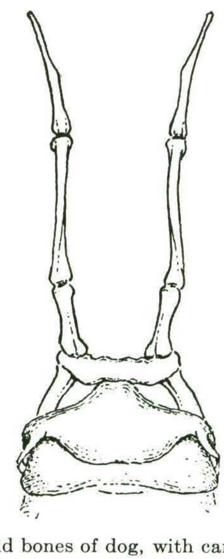

Figure 52. Hyoid bones of dog, with cartilages of larynx.

The male organ of a great many mammals, such as seals, walrus, raccoon, and other carnivores, as well as beaver and many other rodents, is provided with a bone, the os penis, or baculum. In addition to being an integral part of a complete skeleton, this bone in some groups of mammals has diagnostic characters of generic significance. This bone does not articulate with the rest of the skeleton, but has ligamentary attachments, and should always be looked for. When found it should be left attached to the pelvic bones if possible, as it is easily lost.

Cetaceans (Whales, Porpoises, etc.)

The only satisfactory way devised for exhibiting the body appearance of cetaceans is by constructing a model from measurements and casts. Collecting the skeleton except for its size causes little difficulty. The slender cheek bones of porpoises require care, and the pelvic bones or rudimentary hind limbs should always be looked for. The pelvic bones are small and deeply embedded in the flesh, with only very slight ligamentary attachments, so that they are hard to find (Figure 53). In a porpoise 7 or 8 feet long the pelvic bone may be only 2 or 3 inches long. The last rib often lies

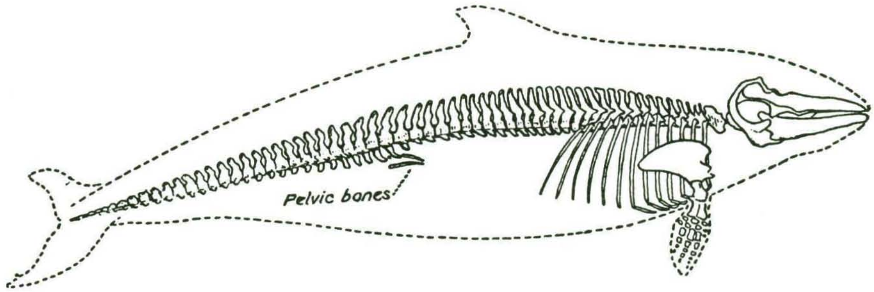

Figure 53. Skeleton of porpoise, showing pelvic bones.

loose in the flesh, with its upper end several inches from the backbone. The chevron bones along the under side of the tail should be saved. There are no bones in the *sides* of the tail or flukes, nor in the back fin, so they may be cut off close to the body and thrown away. The hyoid is usually largely developed, and firmly attached to the base of the skull.

Bird Skeletons

Bird skeletons are a little more difficult to prepare than those of mammals, as the bones are lighter and more easily cut or broken.

The small bones at the tip of the wing should be left untouched, and it is well to leave the outermost wing quills attached to avoid removing any of the small bones with the skin. When ossified tendons are found, as in the leg of the turkey, or on the under side of the neck, they should be left in place. Be careful not to break off the slender bony points on the neck vertebræ, the projections from the rear sides of the ribs (uncinate processes), and the last bone of the tail. In most birds the neck and back can be left untouched, as the muscles will dry up. The hyoid bones, which support the tongue and are attached to the windpipe, as well as the windpipe itself when it has bony rings, should all be kept. As many birds, especially birds of prey, have a ring of bones in the eyeball, it is best to simply puncture the eyeball to drain off the fluid content and leave the eyeball in place. Remove the brain carefully and do not cut the skull in any way, as might be done in making a birdskin. As there are small accessory bones on some birds' skulls, the skull should not be trimmed up too closely. Make the skeleton into a compact bundle for packing by bending the neck back and folding the legs and wings closely alongside the body.

On several occasions when field camps were infested by large numbers of yellow-jacket wasps, which came to carry away bits of meat from the skinning table, it was found that if the skin of a small mammal was roughly removed and the carcass hung up on a string, that the wasps would soon remove virtually all the flesh, sometimes within 15 minutes, leaving a good ligamentary skeleton without disturbing small bones like hyoids, patellæ, etc.

The easiest, and in many ways best, way to collect small bird skeletons is to place the entire bird in alcohol (about 75 per cent strength), first making an incision in the abdomen to allow the alcohol to reach the viscera. Formalin dissolves the mineral matter from the bones if they are left in the solution for any length of time. If nothing else is available, use weak formalin, one part of formalin to twelve of water, and remove the specimen as soon as possible.

Fishes, Reptiles, and Amphibians

Fishes, small reptiles, and amphibians are best treated by preserving them in alcohol. If they are not primarily intended for skeletal material, formalin solution may be used. Large turtles may be opened by sawing through the bridge which, at each side, joins the shell of the back and under surface, and the flesh cut away. As the bones are frequently soft, be careful not to cut into them. Fish may be cut open and the viscera removed. Cut away the skin, and beginning at the lateral line, scrape the flesh from the ribs, working towards the back. Pick away any loose flesh from the base of fins and skull, but leave the gills in place. Brush away blood and mucus with water, and soak the skeleton in clear water for an hour or two. The skeleton may then be preserved in alcohol, or dusted with dry arsenic and hung up by the head to dry. The pectoral fins should be spread alongside the body for protection.

Packing Skeletons

A skeleton, especially a small one, should be thoroughly dried, as a damp specimen may "sweat" and rot the ligaments that hold it together. With a specimen the size of a deer, it will be necessary to disjoint the backbone just behind the ribs in order to make a compact bundle. A larger animal may have the backbone separated into as many sections as convenient and the leg bones separated at each joint. There is really no limit to the subdivision of a large skeleton, as the bones will all be separated ultimately for cleaning. It is well to bear in mind, however, that small pieces are more apt to become lost. An axe or saw should never be used in disjointing skeletons of any size. By paying careful attention to the anatomical construction of the joints, the largest land mammals of North America can be readily taken apart with a skinning knife, without breaking or chipping bones.

If necessary to reduce space, the breast bone may be separated from the ribs by cutting through the cartilage *just below the bony end of each rib.* The breast bone and attached cartilages should be carefully dried and preserved. The ribs can then be detached from the backbone, and when dismantled in this way a good-sized skeleton may be packed in a flour or beef barrel.

Rags, tow, crumpled paper, etc., should be wrapped around the front teeth of deer and similar mammals to prevent the incisors from chipping. In fact, it is advisable to put some loose packing between the jaws of any large mammal. Boxes should be tight to shut out dogs, rats, and mice. Straw or hay is the best packing material, but moss, excelsior, etc., will do, the main point being to prevent the loose bones from rattling about and breaking. Seaweed should never be used for packing, as the salt gathers dampness. A little salt may be sprinkled on bones of a large animal if putrescent. Some aquatic mammals, such as seals and porpoises, can be packed in salt without detriment to their bones, but small skeletons should never be salted. Alum should never be used on a skeleton.

Cleaning Skulls and Other Bones

As cleaning and finishing skulls and skeletons is a more or less technical laboratory process, most museums prefer to receive skulls and bones with merely enough superficial cleaning to prevent them from becoming offensive. Fresh muscular attachments, periosteum, and ligaments are tough, stringy, and hard to scrape off. After these attachments have dried hard they protect the specimen from breakage in shipment, and when softened in water they are easily removed and with less risk of injuring the bony tissue.

The method of cleaning skulls for scientific purposes is, however, relatively simple, and many private collectors will want to clean the mammal skulls in their collections. In some cases it is virtually impossible to determine species without examining skull and tooth characters, and this is hard to do unless the skull is cleaned.

The simplest method of cleaning adult skulls and bones is by maceration, merely placing them in fresh water, in wooden barrels or stone crocks, and allowing them to remain a few weeks or months until the animal matter decays and disappears. This method is slow and causes an offensive smell, but where conditions permit its use, maceration probably gives the most satisfactory results. When carried on away from dwellings accidents may result in loss of the specimens, and when on city roofs the water becomes dirty with soot and discolours the bones.

If only a few skulls are to be cleaned, they may be placed in fresh water and gently boiled until the flesh and ligaments are loose enough to be separated from the bone with a small scalpel or to be picked off with fine-pointed forceps. Be very careful not to scrape too hard and thus scratch the bone, nor to break off thin processes of bone, particularly around the eye or on the base of the skull.

The collector has been warned to remove the brains from the skull in the field, as, if the brains are not removed before cooking, they may expand and force the brain case apart. If the brains are still in place, the skull should be first soaked in warm water and the brains removed with a bent wire, a pinhead, or a small scraper. A small (half-ounce) rubber syringe is very useful: one hand can operate the syringe while the other holds the skull under water. If the brain is thoroughly softened and broken up, the greater part of it can be sucked out with the syringe, instead of being forced out, with no danger of disarticulating the posterior part of the skull.

If it is desired to whiten the skulls, rinse them off after cleaning and let them simmer for a short time with a little hydrogen peroxide in the water. If they are greasy, soak them for a few hours in benzine or carbon tetrachloride. It is not generally advisable to whiten skulls very much, as it is hard to see the sutures in a chalky white skull when studying it.

Several small skulls may be boiled at the same time if adult and young specimens are not mixed up. The juvenile specimens will not stand much boiling without falling to pieces. Put each skull in a small, cylindrical glass vial with label (number) written with pencil on a piece of tough paper or stencilled on pure tin, aluminium, or Monel metal. Put enough water in the vials to cover the skulls, plug the vials with a bunch of cotton, set in a saucepan, and boil as long as necessary. The heat is easier to regulate if a gas-burner is used.

The usual method of mending broken bones is by using a good brand of liquid glue or celluloid dissolved in acetone. The writer has recently examined some small skulls mended by Mr. J. Dewey Soper, using liquid solder in tubes. He finds that if there is no objection to the silvery colour, which is barely noticeable if the joint is neatly made, the solder dries rapidly and offers rapid control and manipulation. The solder is especially useful in uniting the members of separated lower mandibles. Treat the edges of both parts with the solder, bring into contact, and almost immediately the specimen may be laid aside without further holding, and the parts are there to stay.

Holden's Method of Cleaning Skulls. Holden's method (1914, 1917) of cleaning skulls by stewing in a solution of crescylic acid (C_3H_4 CH_4 OH), ammonia, and water was formerly used in the National Museum of Canada, but has been discarded on account of the difficulty of keeping the strength and time of boiling exactly right for specimens of different ages.

Rowley's Method of Cleaning Skulls and Bones. As boiling has a tendency to crack and loosen teeth, Rowley (1925, 214) recommends that skulls be treated separately in the same manner as ligamentary skeletons. The solution used is water, 1 gallon; pancreatin, 2 heaping tablespoonfuls; sodium sulphide, one tablespoonful. The pancreatin acts as a digester in softening flesh. Too much heat is bad; a low, even temperature accompanied by motion in the solution is best, and may be obtained by a steam pipe playing into the solution. A simple apparatus may be made from a gallon oil can. Solder a crossbar to the screw cap so it is easily detachable. Solder a nipple over a small hole on the other side of the top and attach a piece of flexible gas-stove hose or tubing to the nipple. Fill the can three-quarters full of water and set it on a gas plate. When the water boils and the steam comes out of the end of the tubing, lower the gas jet until it just keeps the water boiling, and stick the end of the tubing into the receptacle holding the solution and the bones. A glass preserve jar is good for small specimens, and the temperature of the solution will rise to 208 degrees F. in a quart jar. An open, white-enamelled bowl about 8 inches across, with the same amount of steam, will keep the solution at about 180 degrees F., a good temperature in which to work small skulls and skeletons. Small stone crocks are cheap and excellent for this purpose. A string may be tied to each specimen so that it may be lifted out from time to time and examined. As the steam condenses in the solution the liquid may be dipped out of the vessel, and if necessary more sulphide added to compensate for the dilution. When clean, the bones are placed in warm water for a short time, and then dried, degreased, and bleached. If the bones are not as white as desired, they may again be degreased and bleached.

Cleaning Skulls and Bones with Aid of Dermestid Beetles

The various species of the genus *Dermestes*, known popularly as bacon beetle, buffalo bug, etc., have long been rated as destructive pests that attack drying hides, pelts, dried meats, and other animal products, as well as zoological specimens of all kinds that are not thoroughly poisoned. Collectors have found in the past that certain insect larvæ will clean the animal matter from unpoisoned bones, and if the bones are exposed too long the ligaments are eaten away, small bones fall apart, and becoming mixed are difficult to sort out. C. D. Bunker of the University of Kansas is generally credited with being the originator and pioneer of a practical method of cleaning bones with the aid of insects, and the method has been used successfully by A. Brazier Howell in the Smithsonian Institution, Remington Kellogg and E. R. Kalmback in the U.S. Biological Survey, Harry C. Raven in the American Museum of Natural History, E. Raymond Hall in the Museum of Vertebrate Zoology, and Clifford Hope in the Royal Ontario Museum of Zoology at Toronto.

Messrs. Hall and Ward (1933, pages 359-360) describe the method now used at Berkeley, California, employing dermestid beetles of the species *Dermestes vulpinus*. A sufficient number to start a colony may be found in decaying carcasses of vertebrates, especially in those where some or all of the meat has "dried up". A fair number of adult beetles is required in a working colony for reproductive purposes, but the growing larvæ consume the greater amount of food and are the most useful as bone cleaners. In the Life Sciences Building at Berkeley, a "bug-proof" room 8 by 10 feet is warmed by a steam radiator regulated by a thermostat so as to maintain a constant temperature of 84° Fahrenheit. A pan of water placed on the radiator provides the necessary humidity. The walls have steel shelving that supports wooden boxes 3 feet long, 18 inches wide, and 1 foot deep, in which the beetles are reared and allowed to work. A strip of tin 3 inches wide, laid around the inside of each box at its top, keeps the "bugs" more or less confined. A few adult beetles are put in one of the boxes with a partly dried carcass and left for about a month. When a large number of larvæ are concentrated in a small space, a diminishing food supply forces them to work on any new material introduced. The hard dry specimens received from the field are placed one layer deep on layers of cotton in shallow cardboard boxes with another layer of cotton covering them. These cardboard boxes are then placed in the boxes containing the "bugs". The "bugs" will usually clean small skulls in from 24 to 48 hours, depending upon the number of "bugs" and the amount of food remaining in the carcass used for nursery purposes.

After removing the skulls from the "bug-room" the lower jaw of each is disarticulated and the cranium, jaws, and tag are put in a vial. Ammonia, concentrated, 26 per cent is added, and the vial corked and allowed to stand for 12 hours. The ammonia will loosen any pieces of meat or cartilage overlooked by the "bugs" and will also remove excess grease in the bone. The ammonia is then replaced by water and the skull allowed to soak 24 or more hours, depending on its size. After pouring off the water the skull is washed in a fine stream of warm water with a cross current of compressed air. This can be done in a small sink, preferably set in a work table. Above this, connections to hot and cold water lines empty through an outlet to permit of regulating the temperature of the water discharged. A blowpipe, run through a rubber cork that is inserted in the nozzle of the faucet, provides a stream of small diameter adjustable as to velocity. Another blowpipe, inserted in the end of a rubber hose connected with the compressed air line, is so arranged that its outlet can be brought near the outlet for water. The rinsing in the air and water completes the cleaning operation. The specimens are placed in small pill boxes to dry, and after 24 hours they are put in vials and are ready to be sent to the cataloguing room.

Certain precautions must be taken: (1) the tags must be of parchment or water-resistant fibre composition and the data must be written with ink that will withstand submersion in water and ammonia; (2) skulls that have large porous tympanic bullæ filled with blood, and skulls of very young individuals, should be dry when put in the "bug-room" and should be exposed to a limited number of "bugs".

In conclusion it can be said that by use of the dermestids one worker can clean, thoroughly and without the slightest damage to the most delicate processes, many times the number of specimens he can complete with any other known method. Processes are not broken off; delicate structures are not destroyed; teeth do not fall out; enough non-greasy animal matter remains in the bone to prevent deterioration; and sutures do not gape even in young specimens. All these are advantages over cooking. The preparator using this process may clean up to 100 and even 200 skulls per day. Also, the employment of compressed air as a substitute for instruments is an improvement which can not be too highly recommended.

E. R. Kalmbach, in charge of Food Habits Research Laboratory, Bureau of Biological Survey, Denver, wrote that "We have found that in the case of three of the five workers here a noticeable irritation to membranes of the nasal passage and even the skin when the dust-laden material of our 'bug-house' is being handled. In one instance the irritation took the form of an asthma and caused us to resort to respirators placed over the nose while working with the specimens before they are treated with ammonia and peroxide. It appears that the finely barbed hairs on the larval sheds of these insects causes this irritation . . . . Our work is done entirely with the large forms of the genus *Dermestes*. . . . When the colony is watched so that the normal food supply is not completely consumed, we have had no trouble in this direction (larvæ feeding injuriously on delicate bones), and such bones as the ribs of hummingbirds and the digits in bat wings come through uninjured."

Borell (1938, pages 102-103) states that many collectors have not appreciated the efficiency of dermestids for cleaning small collections of skulls and skeletons when no special facilities are available. While doing field work in Texas a number of dermestes were obtained from an old coyote carcass and the dried feet of a cow. These were put into 2-pound coffee cans and the small mammal skulls on hand were placed in the cans between layers of cotton. As more skulls were acquired they were placed in the cans with additional layers of cotton. When he was ready to leave the field the cans were fitted with light covers and brought to the office by car. Upon arrival the contents of the small cans were transferred to a 3-gallon bread tin with a tight-fitting lid. Such a closed metal container was necessary to prevent the beetles from escaping into the building. Although in a tin can, with little or no fresh air for over 4 months, the beetles are still thriving, and continue to rapidly clean any skull or skeleton, provided, of course, food in the form of dried meat has been furnished when uncleaned skulls or skeletons were not on hand. After taking the bones from the beetle can, they were soaked with ammonia and water essentially as in the method described by Hall and Russell. Upon removal from the water any small bits of tissue remaining were easily separated with a scalpel or brush. Glass jars or clean, new tin cans should be used for soaking bones; otherwise they may be stained. As low temperatures make the beetles and larvæ inactive, and because vibrations are disturbing to them, it is desirable that they be kept in a warm, quiet place.

The M. V. Z. Method of Cleaning Large Skulls and Bones. Although the routine practice at the Museum of Vertebrate Zoology is to use dermestid beetles in removing flesh from small skulls, Hall and Russell (1933) state that experience has shown that it is better to "cook" skeletons as large as deer, moose, and mountain lion instead of "bugging" them.

When skulls are to be cooked, it is best to soak them first for 12 hours in a solution of water and ammonia (proportion, 2 quarts of water to an ounce of ammonia). In cooking, 1 tablespoonful of mild soap flakes and 15 cc. of concentrated ammonia are used to 2 or 3 quarts of water. This solution is brought to near the boiling point and cooked ("simmered", not "boiled") until the meat is soft and transparent. To remove the meat, a fine stream of condensed air is far superior to instruments or water. Also it is more thorough and the loss of lachrymals and zygomatic arches is greatly reduced for the reason that these parts are more easily displaced by touch of an instrument than by the air. The air, strangely enough, does not blow off these fragile bones even when striking them with considerable force. One must be careful, however, to keep a finger over the teeth, for there is danger of blowing them out in the case of those specimens that have been cooked. The cavities of large leg bones are filled with marrow, which must be eliminated as soon as possible to prevent it from soaking into the bones and staining them. If a small hole is drilled in each end of the bones at the time of cooking, the water will loosen the marrow and it can be blown out with compressed air. Even though the skeleton may have been roughed out well, there is enough oil in the remaining meat thoroughly to soak the bone, if it be allowed to remain uncleaned for as long a time as is required for the bugs to clean it. The soaking of large skulls and skeletons in many changes of water will remove the blood. Although it is usually impossible to soak them in the field, opportunity should be taken to soak such specimens as arrive at the laboratory in a fresh condition. Never use water that is hot; this will set the blood in the bone. (In the northern woods and mountains it is usually practicable to put large skulls and bones in gunny sacks and immerse them in a brook or pond over night.)

Degreasing Bones

Degreasing is done by placing the bones in carbon tetrachloride, benzine, or pure gasoline in glass jars and leaving them in the sunshine until the grease is dissolved. If benzine or gasoline is used, a false bottom of wood is fitted into the jar, so that the bones will not lie in the grease that soaks out of them. Benzine and gasoline are inflammable and explosive, and some cheap grades of gasoline contain a gummy substance that will turn bones yellow. Carbon tetrachloride has none of these faults and is safer and preferable for other reasons. It is a colourless compound, only slightly poisonous, and is very heavy, so that grease floats to the surface. If the solutions become dirty, they may be skimmed, filtered, and used again.

Treatment of Teeth

The teeth of large skulls, particularly those of bears, wolves, deer, etc., have an annoying tendency to crack. This is usually due to sudden changes of temperature, which cause the enamel to shrink or expand at a different rate from that of the bony centre of the tooth. Therefore, the skulls should not be suddenly changed from hot to cold water. The cracking of teeth may be diminished to some extent by coating them with melted paraffin, but this method is not very dependable. A much better method is to coat each tooth with a thin solution of Ambroid cement, thinned down with Ambroid solvent or acetone. This forms a very thin elastic film on the tooth and prevents it from cracking, or if the tooth is already cracked, holds the pieces together. If the Ambroid cement is put on too thickly, it discolours the tooth slightly, but it may be dissolved and sponged away with acetone at any time.

A more recent and better method described by Mr. A. Lucas (1924) consists of using celluloid dissolved in acetone. Merely by varying the proportion of acetone in which the celluloid is dissolved the solution may be used for coating teeth, for impregnating porous materials that are in need of strengthening, as a waterproof, air-tight varnish, or as a strong but semi-flexible cement. It is prepared by cutting colourless sheet celluloid into small pieces and dissolving them in a bottle of acetone. If it becomes too thick it should be thinned with acetone; if it is desired as a cement, it may be allowed to evaporate for a few minutes in a shallow dish, when the viscosity will increase rapidly (Leechman, 1931, 131-2).

Teeth, particularly the milk teeth of young animals, frequently drop out in the cleaning process, and care should be taken not to lose any. The ordinary method of setting loose teeth is to dabble a wisp of cotton in liquid glue and wrap it around the root of the tooth before putting the tooth back in the socket. A better method is to force a small pellet of plasticine into the tooth socket. This will hold the tooth effectively and permit its removal for examination at any time without danger of cracking the alveoli of the jawbone.

After the skull is cleaned and bleached, the cranium and lower jaws each have the catalogue number and sex neatly marked on the bone in black carbon ink. Skulls are frequently laid out in series for study and if not plainly marked are apt to become mixed. Small skulls are most conveniently kept in cylindrical glass vials of appropriate size (2 inches depth, and diameter varying from ¼ inch to 1 inch). Larger skulls may be kept in pasteboard boxes. Large skulls should have the jaws separated by a bunch of crumpled paper to avoid chipping of the teeth.

Whether small skulls are kept in the same storage case as the skins to which they belong is a matter of individual preferences or circumstances. Skulls are apt to rattle about when trays are taken out, and in practice skulls and skins are usually compared separately. However, it is often convenient to keep them together, making it easy to take out individual skins and skulls when desired. If kept with the skins, the skull vials or boxes may be laid in narrow pasteboard trays either in the middle or at an end of the sliding drawers. A slight economy in storage space may often be effected by keeping the skulls in separate cases, arranging them in systematic order with the species or subspecies in their numerical order according to the catalogue numbers.

Incisor Teeth of Ruminants

Hollister (1923) calls attention to the fact that even in collections of dozens of skulls of a given species of deer, sheep, antelope, or other ruminants, it is hard to find a perfect set of incisors. The incisors in the lower jaw are very fragile, many are chipped or broken before reaching the museum, and after cleaning and drying in a heated building they become as brittle as delicate china. Being attached to heavy skulls, they are almost sure to strike some hard surface and break. As the incisor teeth are of great scientific value in systematic work, Hollister advocates that every perfect set should be carefully preserved by sawing off the ends of the jaws just posterior to or across the middle of the junction of the jaws (symphysis menti). Number and label the specimens and preserve all the incisor row complete, in natural position, in trays of cotton, like collections of eggs. If it is necessary to photograph a complete jaw, the severed end may be fastened temporarily with modeling clay or plasticine.

PERMITS FOR SCIENTIFIC PURPOSES

The greater number of migratory species of birds are protected throughout the year by the Regulations for the Protection of Migratory Birds, adopted under authority of the Migratory Birds Convention Act (R.S.C. 1927, Chap. 130, as amended by 23-24 Geo. V, Chap. 16, 1933), which is a Dominion Statute. Permits for collecting or banding migratory birds in Canada are issued by the Chief, Canadian Wildlife Service, Department of Northern Affairs and National Resources, Ottawa, under the following regulations:

> "Migratory game, migratory insectivorous or migratory non-game birds or parts thereof or their eggs or nests may be taken, bought, sold, shipped, transported or possessed for scientific purposes, and said birds may be captured for banding purposes, but only on the issue of a Permit by the Minister or by any person duly authorized by him.
>
> "Such Permits may, upon application, be granted to recognized museums or scientific societies, and to any person furnishing written testimonials from two well-known ornithologists, or at the request of any Department of the Government of Canada, the United States of America, or any other national government, or any Department of the Government of any Province of Canada or any one of the States of the United States of America.
>
> "The return of specimens taken under such permits shall be made to the Minister upon the expiration of the Permit."

Mammals are generally resident, or non-migratory, and the control of mammal life in Canada, except in the Northwest Territories and the National Parks, is vested in the provinces. Most of the smaller species, such as mice, shrews, moles, gophers, chipmunks, etc., are not protected. Permits to collect game mammals and certain species of fur-bearing mammals must be obtained from the Game Departments of the appropriate province or territory. Applications for permits to collect in the Northwest Territories should be sent to the Commissioner, Northwest Territories, Ottawa, and in Yukon Territory to the Commissioner, Yukon Territory, Northern Affairs and National Resources, Whitehorse, Y.T.

Export of wild birds or mammals or parts thereof from any province is controlled by the Game Export Act (4-5 Geo. VI, Chap. 17). Export of furs from the Northwest Territories is subject to the Fur Export Ordinances (Chap. 41, Revised Ordinances, N.W.T., 1956). Export of furs from Yukon Territory is subject to the Fur Export Ordinance (Revised Ordinance, 1958).

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"The district covered is, approximately, the northeastern United States and southern Canada, extending from Labrador westward to and including Manitoba and North Dakota, and southward and including North Carolina and Kansas." (In bringing nomenclature and descriptions up to date the author acknowledges help of Dr. Hartley H. T. Jackson on the mammals, Dr. Carl L. Hubbs on the fishes, Dr. Emmett R. Dunn on amphibians, Dr. Alexander G. Ruthven on reptiles, and Dr. H. C. Oberholser on birds.)

Since the first appearance of this work in 1876 up to the thirteenth edition in 1929, it has been generally admitted to be the most concise and practical handbook for identifying both land and water vertebrates found in this region. The keys are workable and up to date, the etymology of scientific names is interesting and useful, and a large number of modern naturalists began with it as schoolboys and have kept it on their desks ever since.

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