Stansbery
1992

THE OHIO STATE UNIVERSITY

Museum of Zoology

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6 October 1992

Dr. Richard Neves
Virginia Coop. Fishery Unit
Virginia Polytechnic Institute and State University
Cheatham Hall
Blacksburg, VA

Dear Dr. Neves:

I have gathered together everything I could find pertaining to processing bivalve mollusk specimens here at OSUM. Most of these items were written some years ago, to help in training student employees (back when we had them, before "trickle-down" economics), and all were written before we computerized; so some labeling and record-keeping procedures have changed since then. Still, I hope this is of some help. If you (or Dr. Hoffman) have any questions, just let us know.

Sincerely,

Kathy G. Borror
Curatorial Assistant
Bivalve Mollusk Division

cc: Dr. David H. Stansbery

MOLLUSK COLLECTIONS AT THE OHIO STATE UNIVERSITY MUSEUM OF ZOOLOGY

David H. Stansbery
Curator of Bivalve Mollusks

and

Carol B. Stein
Curator of Gastropod Mollusks

The Ohio State University Museum of Zoology
1813 North High Street
Columbus, Ohio 43210

October 1983

Reprinted from: Miller, Andrew C., Compiler. 1983. Report of Freshwater Mussels Workshop 26-27 October 1982. Published by U.S. Army Engineer Waterways Experiment Station Environmental Laboratory, Vicksburg, Mississippi 39180

MOLLUSK COLLECTIONS AT THE OHIO STATE UNIVERSITY MUSEUM OF ZOOLOGY

by

David H. Stansbery and Carol B. Stein*

In the early and middle 1800's a gentleman named Henry Moores collected mollusks in central Ohio. He exchanged many of his specimens with Thomas Say and Isaac Lea of the Academy of Natural Sciences of Philadelphia, with John Gould Anthony of the Museum of Comparative Zoology at Harvard University, with an unknown correspondent at the Smithsonian Institution, and with many other early conchologists.

Henry Moores' collection was eventually purchased by The Ohio State University, where it became the nucleus of the University's first Museum of Zoology in the late 1800's. But interest in the museum's shells waned during the mid-1900's, and so far as we can determine, the collection was not used for research for many years. The shells lay unused and all-but-forgotten in open trays in attics and behind exhibits. They became covered with thick layers of soot from the soft coal that heated Columbus buildings before natural gas was used.

In the early 1960's we found both a challenge and an education in cleaning the shells that had been collected a century earlier. In the process, we could not help but observe how different these specimens were from those being currently collected. Growth rates were, for example, frequently much greater in the 1960's than a century earlier. We related this to the increased use of fertilizers, but objective evidence is lacking.

The Henry Moores collections now form a relatively small part of the extensive collections of bivalve and gastropod mollusks at The Ohio State University Museum of Zoology. Over the past two decades these collections have grown at a remarkable rate. Our staff and students have collected hundreds of thousands of freshwater mollusks in many of the streams of North America. Our unionid and pleurocerid collections are among the largest in existence. We have built unusually extensive collections of soft parts of these mollusks. The many large series of specimens have a high degree of statistical significance, making them especially valuable for research involving variation within and between populations. We have over 50,000 lots of unionids alone, which may be larger than any other recent collection of these animals in the United States.

Since such a high percentage of the specimens in the OSUM collections have been collected within the past quarter-century, this is not necessarily the best place to study the early distribution of mollusks. To examine type specimens and other historical material, one should visit the United States Museum of Natural History in Washington, D. C., the Museum of Comparative

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* Curators, respectively, of Bivalve Mollusks and Gastropods, Museum of Zoology, The Ohio State University, Columbus, Ohio.

Zoology at Harvard University, the University of Michigan Museum of Zoology, or the Academy of Natural Sciences of Philadelphia.

Some visitors, upon seeing the OSUM collections of freshwater mollusks, remark that now they understand why these animals are endangered! Actually, many of our specimens are shells obtained from midden left by muskrats and other mollusk-eating animals. Sometimes these shell middens are quite extensive. We once filled 18 three-gallon buckets with nested empty fresh unionid shells from a single large midden.

The collections have also grown through the generosity of others. Small but valuable collections are often donated by schools, colleges, and individuals who no longer require research collections. Specimens brought in for identification are frequently added to the collections, supplementing our own collecting and providing samples from sites we have not been able to visit.

Much of our mollusk collecting is done by hand in shallow water in small streams and along the shores and shoals of larger rivers. The museum also owns a large jon boat, which we use for collecting in medium-to-large rivers where SCUBA or crowfoot brailing are more effective collecting methods. The boat and motor are pulled to the field sites on a trailer. Our field laboratory is a modified camping trailer.

The Museum is located on the lower floor of Sullivant Hall at the main entrance to the University campus in Columbus. One of the first things visitors notice when they enter the Museum is our large collection of maps (Fig. 1*). The walls of the main hallway are nearly covered with those maps we most often use for quick reference. We also have an extensive map library (Fig. 2) which contains road maps, books of county maps, state and national drainage maps, and most sizes of U. S. Geological Survey topographic maps, as well as gazetteers. We are especially eager to obtain and preserve old maps and gazetteers, since these include old place names which "live on" in the literature and on old specimen labels, though they have often vanished from modern maps either because the names have changed or the places no longer exist. The maps are kept in a variety of map cabinets which were all obtained from various salvage or secondhand stores. They have been painted to match, and we have repaired many of them and replaced the handles to make them serviceable. Mr. Charles "Hank" Dowdy, a retired science teacher, is curator of our map library. He is one of several volunteers who contribute their time and effort to help with the seemingly endless work that goes into building a research museum.

The map library exists because of our concern that the best possible locale data accompany all specimens in the Museum's collections. The locales given on the labels of all incoming collections are checked out on detailed maps. If we cannot precisely pinpoint the locality, we contact the collector for more information. For this reason the collector's name is one of the most important items on any field label, along with the exact locale and date of collection. Any information added to that on the original label is placed inside brackets [thus].

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* Figures 1-23 can be found on pages 102-113 at the end of this paper.

Each collection which comes into the museum is given its own number. This number consists of three parts. First is the Museum's abbreviation, OSUM. Second is the year in which the collection was originally made in the field. The third part is the number assigned to that particular collection in that year's series. The three parts are separated by colons. The first collection received at the Museum which was collected in 1982, for example, would be assigned the collection number OSUM:1982:1. If we receive a 1982 collection ten years from today, it will be assigned the next available number in the 1982 series. A field sheet (Fig. 3) is made out for each of these collections, and the sheets are filed by year of collection and then by number. We also enter the field number on a geographic card, so that we can quickly find out what collections we have in the museum from any one locality, such as the Clinch River at Kyles Ford, for example.

Our field labels (Fig. 4), which we supply to anyone collecting for the museum, contain blanks for all of the detailed locality information which we hope to keep with the specimens. This includes the specific stream or other locality; the drainage, distance and direction from the centers of two towns; section, township and range numbers; and township, county, state, and county names. We now add latitude and longitude, down to the second, to improve our precision and to add a set of permanent, worldwide coordinates. Space is left for remarks so that the collector can note any unusual or significant items which might be of value in future studies.

We acquire collections of specimens faster than they can be processed. A large area in each collection range is devoted to partially processed collections arranged by their collection numbers. These are processed as the time and funds become available. Each is well labeled, and the wet specimens are placed in fresh preservative in rubber-gasketed clamp-top jars. If necessary, the specimens could remain in this stage for a century or more until they can be fully processed into the collection.

Frequently, we receive inquiries as to whether or not endangered mollusk species exist in a particular area. The first thing we do is to check the geographic card file to find out what collections we have from the area. Then we look on the field sheets to find out what has been cataloged from the area, and we check the records for species considered to be endangered. If we do not find records of the species in question, we can go to the uncataloged collections from that locality and examine them for the presence of the endangered species. Even though the uncataloged material is not cleaned, culled, or sorted by species, it is still available for research purposes. It is much easier to unpack a few cartons and jars of uncataloged material than to organize an expedition to go in search of it--especially in midwinter!

The cataloging and processing of a collection of mollusks is very time-consuming. The first thing we do is to rough-sort the material by species. Then we cull the specimens of each species. We try to keep what we believe to be a statistically significant series, generally 30 or more specimens representing both sexes and the entire range of shell sizes present in the sample, as well as a representative sample of specimens preserved in AGW (80% ethanol, 15% water, 5% glycerin). Excess specimens which are in good enough condition for research or study are set aside as duplicate material, and specimens which are of no value are discarded.

Anyone who has a need for examples of a particular species of unionid, or for samples from a specific area, is welcome to write us inquiring about the availability of the desired material. Our duplicate material obviously includes more examples of common species than of rare ones.

After the collection has been culled, the specimens must be cleaned. We attempt to remove all of the environment from the surface of the shell without actually damaging or removing any part of the specimen itself. We have found that it is important to be very particular about cleaning the shells. Sometimes a specimen originally thought to be one species reveals characteristics, when clean, which show it to be a different species. Texture, color, ray pattern, muscle scars, and other important taxonomic characters can be seen completely on a clean shell, but are frequently obscured if the specimen is not properly cleaned.

To clean unionid and gastropod shells we use toothbrushes or handbrushes of varying degrees of hardness, in combination with ordinary household cleansing powder (Fig. 5). Various dental tools have also proven useful for removing foreign materials from shells.

To remove resistant deposits from unionid specimens, we use a Dremel Moto-Flex Tool, Model 232, having a variable-speed control and a flexible shaft with a rotary brush on the end of the shaft. We recommend a brush with stiff nonmetallic fibers similar to toothbrush fibers. Our most experienced processors often use a steel brush on shells with an especially tenacious coating. They must be very careful and use a light touch with this tool, however, since the steel has a hardness of five and the shells are much softer, with a hardness of only three. The periostracum is even softer and is very easily scratched with the steel brush.

Small, fragile gastropod and sphaeriid shells are often cleaned with a camel's hair brush, sometimes under a dissecting microscope. A Bransonic 12 ultrasonic cleaner with a detergent solution is also helpful in cleaning very small shells. Shell cleaning is an exacting task, and it takes a long time to learn to do it well.

After the shells are cleaned, the tentative identifications are verified, and the collection is cataloged. All of the specimens of a given species from a given field collection are considered a "lot," and each lot is assigned its own catalog number. Our catalogs (Fig. 6) are bound "blank books" made especially for OSUM. They have 100% rag paper printed with a line for each lot, and columns for the catalog number, scientific name (which includes the author and date), the initials of the person who identified the specimens, the number of wet and dry specimens in the lot, the field collection number, the name(s) of the collector(s), the specific locality where the collection was made, the township, the county, the state or nation (if not USA), and the date of collection. All entries are made as legibly as possible, and only waterproof, permanent, high-carbon ink is used.

Part of each catalog page is reserved for additional comments and possible changes. For example, as we learn more about the systematics of these animals, our studies sometimes reveal that what was once thought to be a single homogeneous species is actually a composite of two or more taxa. In such a

case, we can go back to the catalog, note the change at the bottom of the page, and remove the specimens of the newly recognized species to a separate lot with a new catalog number.

We catalog all of the lots from a given field collection together, whenever possible, since this saves a lot of writing--we can then use ditto marks for the collections data--and it also lets us see at a glance the faunal composition of the bivalve or the gastropod fauna at that site at that time. The complete list of species and their catalog numbers are then typed on the back of the field sheet (Fig. 7) so we can quickly determine what specimens have been cataloged from that locality.

For each lot, a collection record card (Fig. 8) is typed. These cards are filed in taxonomic order, and within each taxon by drainage order. Simply by looking through the card file for a particular species, a researcher can quickly obtain all of the available recorded data for all the cataloged OSUM specimens of that taxon.

Eventually all of these data will be entered into an electronic data bank so that the information can be retrieved quickly and efficiently by whatever category we wish. Right now we are exploring several alternatives to find the best way to program these data for most efficient use in research.

When catalog numbers have been assigned to the various lots, and the shells have been cleaned, the dry shells are numbered (Fig. 9). In the gastropod collections, some of the shells are so small that it is impossible to write a five digit number on the shell, so specimens less than half an inch long are generally placed in a vial or a gelatin capsule with a slip of paper bearing the catalog number. However, the larger shells are numbered with permanent black or white ink, whichever shows up best on the periostracum.

In the bivalve collection, a more complex system is used. The catalog number, plus a decimal number, is written in permanent black ink on the nacre of each valve of every dry shell in the cataloged collection (Fig. 10). Each specimen in the lot is assigned its own decimal number, beginning with 0.1 for the smallest specimen, 0.2 for the next largest, and so on. This allows one to quickly match up the pairs of valves if they should become jumbled and also allows the researcher to refer to any particular specimen in a lot in a publication if necessary.

The numbered shells of most bivalves are then dipped (Fig. 11) into a solution consisting of 1/4 pound of paraffin dissolved in one gallon of xylene. They are left in a vented hood to dry. As the xylene evaporates, a thin coating of paraffin is left on the entire surface of the shell. This greatly reduces the amount of periostracal flaking and cracking, and yet does not significantly alter the natural color, texture, or appearance of the shell. We do not dip gastropod shells, since very few of them seem to be subject to the flaking of periostracum which is so common in bivalves. Nor do we dip the smaller spheriid species, since under high magnification the paraffin coating is visible.

Each lot of shells is placed in its own tray, lidded box or vial (Fig. 12), together with its label. In the gastropod and sphaeriid collections, small

lots are places in an eight-dram shell vial. A 100% rag content label (Fig. 13) bearing all the data given in the catalog is placed in the vial against the glass, with the left side of the label toward the closed end of the vial. The shells are then placed at the bottom of the shell vial. The vial is filled with a plug of cotton, which holds the shells gently but firmly in place at the bottom and the label firmly against the glass side of the vial. We have found that vials smaller than the eight-dram size do not provide enough space for labels with the amount of data we believe is necessary to keep with the specimens, so the vial is sized to fit the label, not the specimens. Large lots of small snails or sphaeriids which do not fit into the eight-dram vials are kept in boxes with plastic lids. The smallest of these are hinged-lid boxes made completely of plastic. The larger ones are modular pasteboard boxes with lift-off acetate lids, somewhat like Christmas card boxes, which keep the shells from accidentally being jostled from one box to another. Large gastropods are housed like the unionids in open trays in drawers.

Small lots of unionids are stored in open trays (Fig. 14) with special standup labels (Fig. 13) made of durable heavyweight juteboard or 100% rag card stock, as shown in the accompanying illustrations. A piece of thin plastic sponge is placed in the bottom of each tray. This keeps the shells from sliding and bumping into each other when the tray is moved and hence prevents much damage from chipping and cracking of the thin shell margins. The shells gain an extra measure of protection from their positions in the trays. The left valve is first placed nacre-down on the sponge liner, and the right valve is then placed on top of it, also nacre-down. This keeps the shells from rolling about, as they would if the valves were placed together as they are in life. All the valves are placed with the ventral margins toward the front edge of the tray. They are arranged in size sequence in the tray. The standup label is always placed at the left rear corner of the tray and is folded so that all of the collection data can be read without touching either specimens or label. All original labels are kept in the trays with the specimens, along with the OSUM label. The OSUM labels are printed in long strips for ease in typing on our 17-pitch label typewriters (Fig. 15), using carbon ribbon for permanence. They are cut apart after typing, and the standup labels are then folded by hand along a scored line.

Because of the way the specimens and labels are arranged, an investigator can open a drawer (Figs. 16, 17) and scan over the rows of shells and labels, frequently obtaining all the data he needs almost instantly. Geographic arrangement of the lots by drainage system within the species drawers also is an aid to research.

Most lots of shells are kept in drawers which fit interchangeably into strong, relatively inexpensive plywood cabinets (Figs. 18, 19). Labels on the outside of each cabinet identify the taxa kept inside and in many cases also note which drainage systems are represented in that cabinet.

Large series of bivalve shells are kept in labeled boxes on steel shelving (Fig. 20). This material is arranged in the same linear systematic sequence that is used in the cabinets. The catalog number is placed on the end of every box, and each lot also has a locale label on the outside of the first box, as well as a complete typed label inside it. Inside each box (Fig. 21),

the shells are arranged just as they are in the cabinet trays. Two carefully crumpled sheets of newspaper in the top of each box hold the shells firmly in place when the box is closed and tied. A single lot may occupy only one such box, or may be so large that several boxes are required to hold it. We feel that this is a very practical and inexpensive way to store the large lots, and it surely does save on costly cabinet space. Any one specimen of the half-million or so unionids stored here can be located in less than ten minutes.

Nearly all of the mollusks which we collect alive are preserved with their soft parts intact. If time and facilities permit, we relax the aquatic animals in a menthol-water or other solution before fixing them, and the land snails are drowned in air-free jars of water. Unionid shells must be carefully opened a few millimeters and kept apart with a rubber or cork stopper (even a twig or pebble will do in an emergency) before they are carefully placed, aperture ends up, into the AGW preservative. We have found that most sphaeriids and freshwater snails can be preserved adequately, even for micro-anatomical studies, by simply dropping them into AGW. When the field collections are brought back to the Museum, we change them into fresh AGW, removing the stoppers as we go. Inexpensive screw-top, wide-mouth glass jars are used as field containers, but these are not suitable for long-term storage of specimens because most are not tight enough to prevent the alcohol from evaporating. New plastic lid gaskets may solve this problem.

The old-fashioned bail-top glass canning jars with rubber gaskets are excellent containers for alcohol-preserved specimens, but are virtually impossible to obtain now. We currently use two types of jars for most of our wet specimens. For smaller bivalves and for small lots of snails we use glass jars with translucent plastic snap-on lids. These are manufactured by the Wheaton Company and come in 2-, 4-, and 6-ounce sizes. Most bivalves and larger lots of gastropods are kept in rubber-gasketed clamp-top glass jars (Fig. 22) which are made in Europe. Several years ago we joined two other museums in importing a truckload of these jars from France. Since this supply is now running out, we are looking for an economical source within this country. These jars can be quickly opened and closed and are air-tight.

Very large unionid specimens and very large lots of wet specimens will not fit into these jars, since the largest is only a three-liter container. We have used some very large screw-top jars successfully by "buttering" the inside threads of the lids with a melted-together mixture of half beeswax and half petroleum jelly. This compound provides an alcohol-resistant seal, serves as a lid lubricant, and inhibits rusting. This method is time-consuming and messy, however, and the compound must be applied to a dry lid and jar to insure a perfect seal. We are considering using translucent plastic buckets and lids for storage of large series of specimens.

A tape-writer label of stainless steel or plastic bearing the OSUM catalog number is placed into each jar of preserved specimens, in addition to the typed paper label. This is simply insurance against the slight possibility that the label paper may disintegrate or the ink fade.

The specimen jars are stored in a linear systematic sequence on steel shelving. For convenience in organizing and handling the wet collection, we use sturdy, labeled cardboard trays (Fig. 23) with a fire-, alcohol-, and water-resistant coating to hold the jars.

We have found that shells with their soft parts intact kept for many years in AGW do not have the same appearance as shells which are stored dry. In liquid, the periostracum typically becomes darker and pinhead-sized bubbles sometimes form between the underlying prismatic layer and the periostracum. Since the shells store better as dry specimens, we are dissecting out the soft parts of the bivalves, when time permits, and of small lots of snails as they are processed. The soft bodies of the mollusks are placed in individually labeled containers (zipper closure plastic envelopes, vials, or capsules) along with their catalog numbers and stored together in the same jar, while the shells are processed into the dry collection. Since each part of each specimen is marked with that individual's own catalog number, the bodies and shells can be re-associated in the future as they are studied. When operculate snails are dissected and preserved separately, each operculum is placed inside its own shell and held in place by a plug of cotton.

It is impossible to go back in time and take a duplicate sample from a population. Populations are continually changing, in some instances to the point of extirpation or extinction. Environmental conditions, the active agents of selection, are forever changing. Therefore, each museum lot is unique and irreplaceable. Our efforts are directed toward building a collection which will continue to be of genuine research value to scholars long into the indefinite future.

Acknowledgements

We wish to recognize the very real contribution made to this paper by the curatorial assistants of the two mollusk divisions. Both Kathy G. Borror, Bivalve Mollusk Division, and William N. Kasson, Gastropod Division, are intimately familiar with the collections and activities described here by virtue of some years of daily hands-on experience. Their suggestions for improvements in both factual expression and ease of communication were gratefully received.

All of the photographs were taken, developed, and printed by A. E. Spreitzer with the perfectionism which is becoming his trademark.

Kathy Newman proofread and typed the final manuscript with all of the enthusiasm of a budding malacologist.

(This and all succeeding photographs in this paper taken by A. E. Spreitzer)

Figure 1. Frequently used geologic maps and drainage maps line the main corridor of The Ohio State University Museum of Zoology

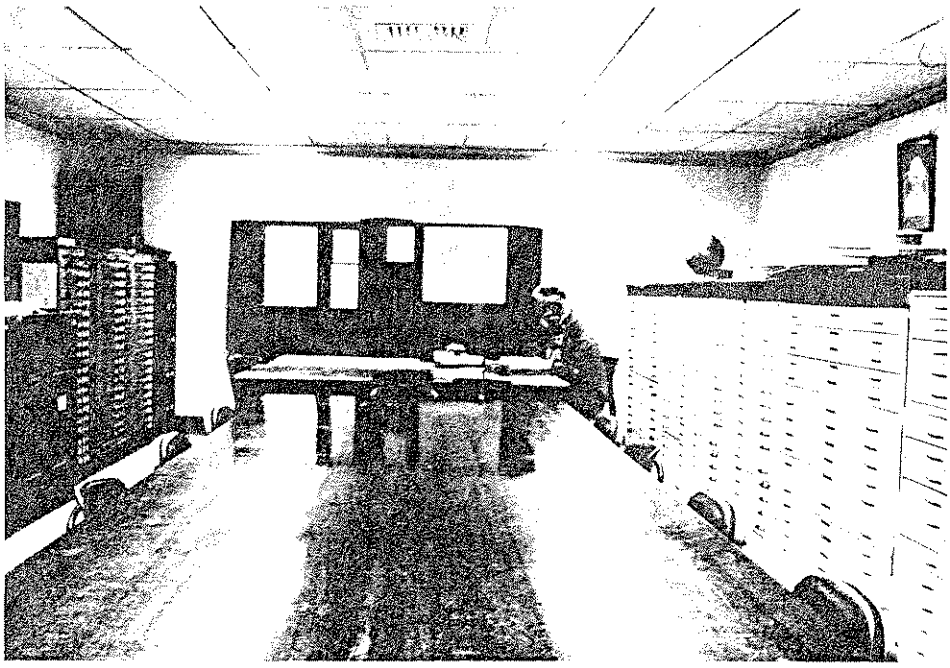
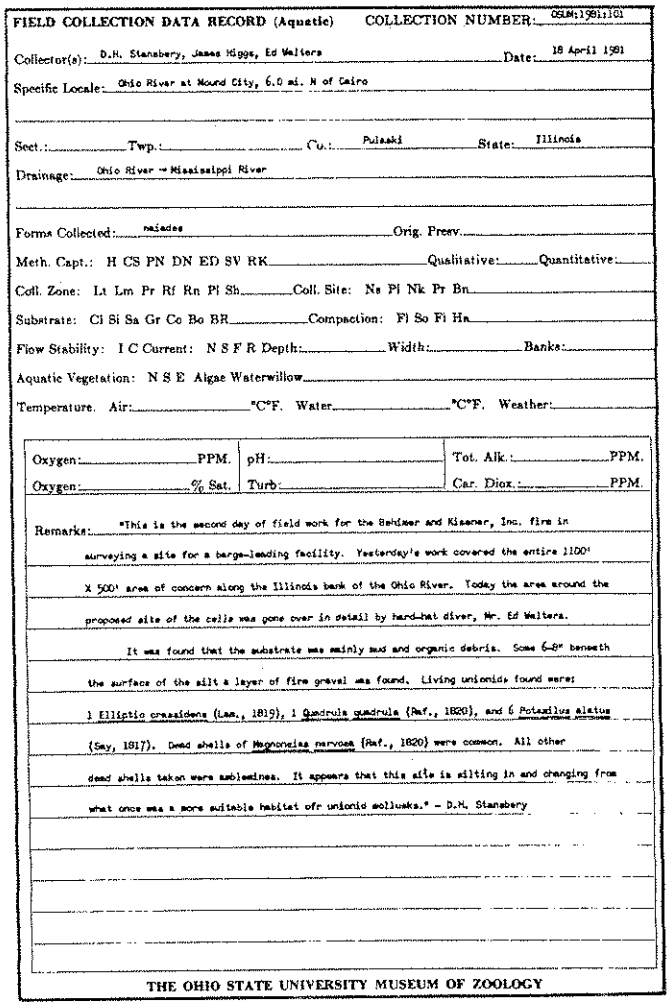


Figure 2. Charles T. "Hank" Dowdy, volunteer map librarian, keeps the OSUM map collection properly organized so researchers can check locales where specimens were collected. The cabinets on the right contain 7-1/2-min topographic maps. Other series of maps and gazetteers are stored along the opposite wall. The large table in the center is used for laying out maps for study



FIELD COLLECTION DATA RECORD (Aquatic) COLLECTION NUMBER: OSUM:1981:101

Collector(s): D.H. Stansbery, James Higgs, Ed Walters Date: 18 April 1981

Specific Locale: Ohio River at Mound City, 6.0 mi. N of Cairo

Sect.: Twp.: Co.: Pulaski State: Illinois

Drainage: Ohio River → Mississippi River

Forms Collected: naiades Orig. Presv.

Meth. Capt.: H CS PN DN ED SV RK Qualitative: Quantitative:

Coll. Zone: Lt Lm Pr Rf Rn Pl Sh Coll. Site: Ns Pl Nk Pr Bn

Substrate: Cl Si Sa Gr Co Bo BR Compaction: Fl So Fi Ha

Flow Stability: I C Current: N S F R Depth: Width: Banks:

Aquatic Vegetation: N S E Algae Waterwillow

Temperature. Air: °C°F. Water °C°F. Weather:

Oxygen: PPM.	pH:	Tot. Alk.: PPM.
Oxygen: % Sat.	Turb:	Car. Diox.: PPM.

Remarks: "This is the second day of field work for the Bechtel and Kissener, Inc. firm in surveying a site for a barge-loading facility. Yesterday's work covered the entire 1100' X 500' area of concern along the Illinois bank of the Ohio River. Today the area around the proposed site of the cells was gone over in detail by hard-hat diver, Mr. Ed Walters.

It was found that the substrate was mainly mud and organic debris. Some 6-8" beneath the surface of the silt a layer of fine gravel was found. Living unionids found were: 1 Elliptio crassidens (Lam., 1819), 1 Quadrula quadrula (Raf., 1820), and 6 Potamilus alatus (Say, 1817). Dead shells of Magnonaias nervosa (Raf., 1820) were common. All other dead shells taken were asiatenses. It appears that this site is silting in and changing from what once was a more suitable habitat for unionid molluscs." - D.H. Stansbery

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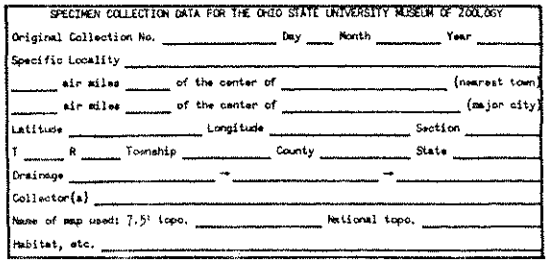
Figure 3. Field sheets such as this are filled out for every incoming bivalve collection, even if the collection contains only a single specimen. It provides a useful place to store a variety of data and remarks about the collection which cannot be put on every lot label. A geographic card file provides a quick means of locating all of the field sheets from any locality or drainage system



SPECIMEN COLLECTION DATA FOR THE OHIO STATE UNIVERSITY MUSEUM OF ZOOLOGY

Original Collection No. _____ Day ___ Month _____ Year _____

Specific Locality _____

_____ air miles _____ of the center of _____ (nearest town)

_____ air miles _____ of the center of _____ (major city)

Latitude _____ Longitude _____ Section _____

T _____ R _____ Township _____ County _____ State _____

Drainage _____ → _____ → _____

Collector(s) _____

Name of map used: 7.5' topo. _____ National topo. _____

Habitat, etc. _____

Figure 4. Field labels such as this one are given to people who plan to collect mollusk specimens for the OSUM collections. Like all OSUM forms, this is made of durable rag-content paper which has great longevity, even when placed in liquid preservative. When such a label is completely filled out by the collector, processing the collection can proceed smoothly when it arrives at the Museum

Figure 5. Using a toothbrush and cleansing powder, Sam Fitton cleans a unionid shell. The moto-tool hanging at the top of the rack is used on some specimens with especially tenacious coatings

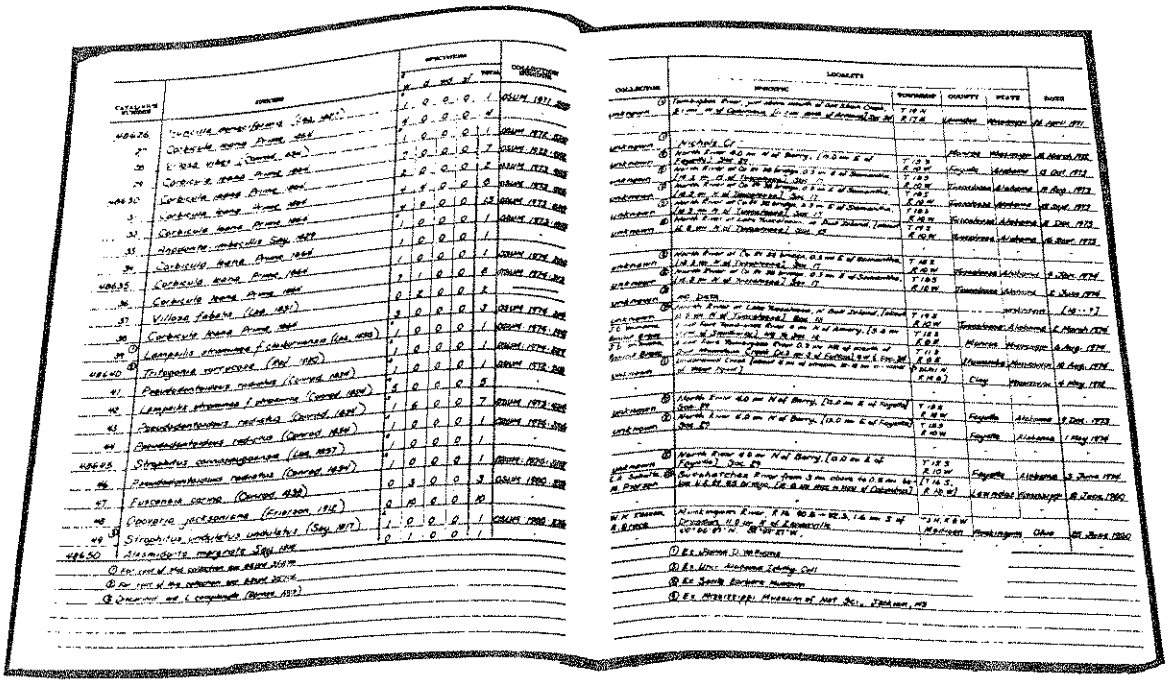


Figure 6. Every lot of mollusks processed into the OSUM collections is assigned its own catalog number and is entered in this permanent bound catalog book. All of the data which will later be typed on the cards and labels are first entered in this book, using permanent waterproof black ink. Each lot has its own line, and notes which pertain to the lots are entered as footnotes at the bottom of the pages as necessary

FIELD COLLECTION DATA RECORD (Aquatic) COLLECTION NUMBER: OSUM:1981:101

Collector (s): D.H. Stansbery, James Higgs, Ed Walters Date: 18 April 1981

Specific Locale: Ohio River at Mound City, 6.0 mi. N of Cairo

Sect: Twp.: Co.: Pulaski State: Illinois

	CATALOG NUMBER	TOTAL NO. SPEC.	SPECIES	I LIVE	II d	II DEAD wd	? SUB-FOSSIL	REMARKS
1	49201	3	Magnonaias nervosa	0	0	3	0	
2	49202	2	Tritogonia verrucosa	0	0	2	0	
3	49203	1	Quadrula quadrula	0	1	0	0	
4	49204	1/2	Quadrula pustulosa pustulosa	0	0	1/2	0	
5	49209	2	Amblema plicata plicata	0	0	2	0	
6	49206	2	Fusconaia ebena	0	0	2	0	
7	49207	1	Fusconaia flava	0	0	1	0	
8	49208	2	Elliptio crassidens crassidens	0	1	1	0	
9	49209	1	Leptodea fragilis	0	1	0	0	
10	49210	8	Potamilus alatus	0	8	0	0	
11	49211	7	Corbicula leana	1	1	5	0	

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Figure 7. On the back of each field sheet is a list of all bivalve specimens cataloged from that collection, if the collection has been processed

COLLECTION RECORD CARD

W	D	WD	SF	TOTAL

SPECIES

RECORDED AS

SPECIFIC LOCALE

TWP. CO. STATE

LAT.-LONG REMARKS

DRAINAGE

COLLECTED BY DATE

IDENTIFIED BY DATE

MUSEUM AUTHOR

CATALOG NO. DATE

COLLECTION NO. PUBLICATION

THE OHIO STATE UNIVERSITY MUSEUM OF ZOOLOGY

COLLECTION RECORD CARD

I	II	♀	♂	TOTAL
LS	SP	DS	SF	
	0	1/2	0	1/2

SPECIES Epioblasma torulosa rangiana (Lea, 1839).

RECORDED AS

SPECIFIC LOCALE French Creek at U.S. Rt. 6 and 19 bridge, at NE edge of Venango, 9.6 mi. NNE of Meadville

TWP. Venango/Cambridge CO. Crawford STATE Pennsylvania

DRAINAGE French Creek → Allegheny River → Ohio River → Mississippi River

COLLECTED BY J.J. and C.S. Jenkinson DATE 9 Sept. 1977

IDENTIFIED BY D.H. Stansbery DATE Oct. 1977

MUSEUM OSUM AUTHOR

CATALOG NO. 40548 DATE

COLLECTION NO. OSUM:1977:176 PUBLICATION

REMARKS

Figure 8. Collection record cards such as this are typed up for each lot of bivalves cataloged into the OSUM collection. They are filed in taxonomic order. The Gastropod Division does not use these cards. Eventually electronic data processing will be used to provide ready access to these data

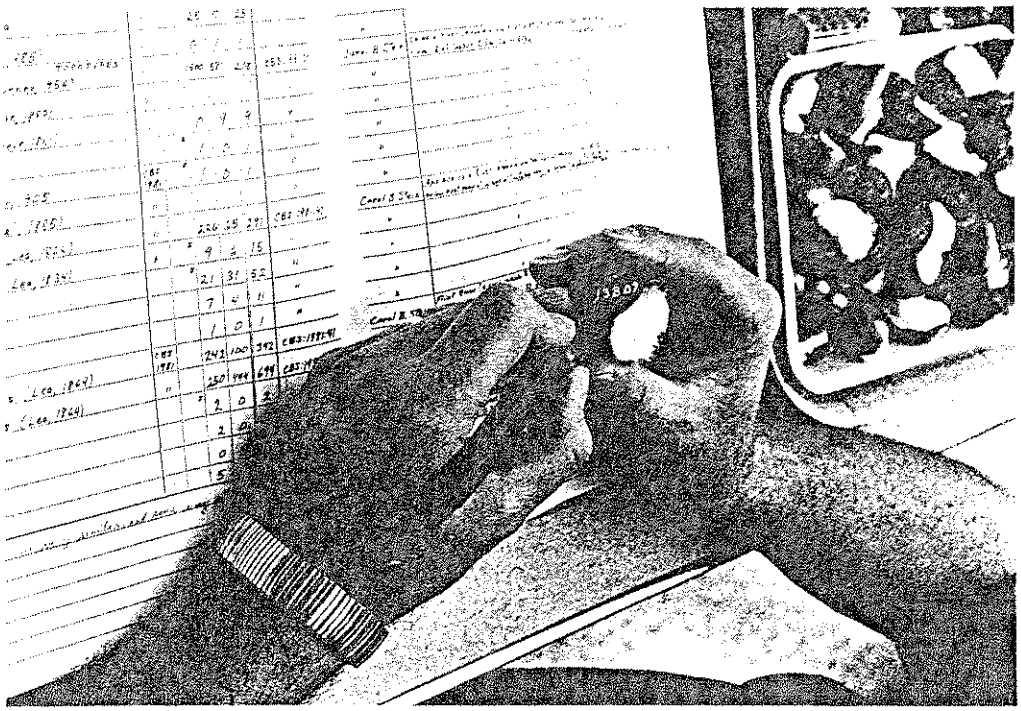


Figure 9. William N. Kasson, curatorial assistant, uses white waterproof ink to number a dark-colored gastropod shell. On light-colored shells, black waterproof ink is used. The soft parts of these specimens have been preserved separately in AGW. The cotton in the aperture of each shell holds the specimen's operculum inside the shell

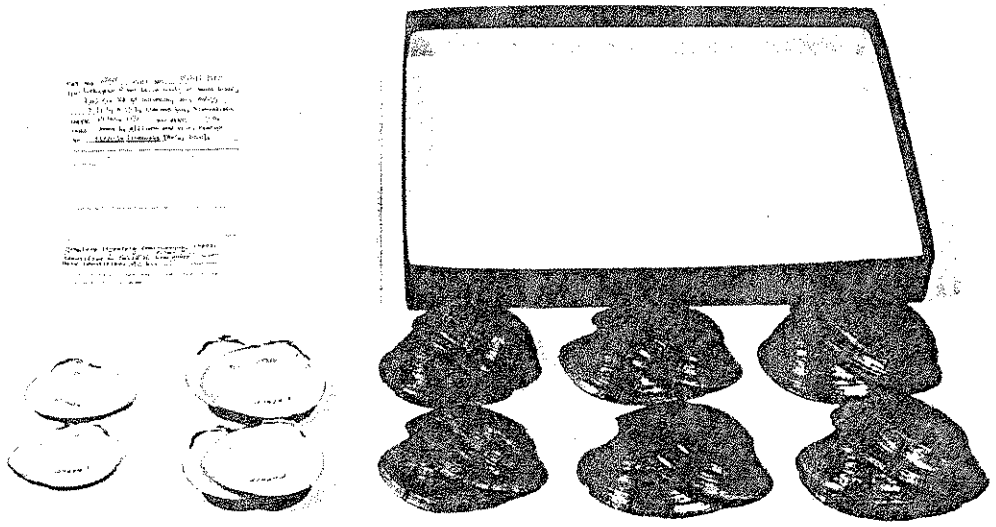


Figure 10. Each valve of every dry bivalve shell is numbered with permanent waterproof ink just inside the pallial line on the nacreous surface. These shells are ready to be placed, together with their original label, OSUM typed standup label, and plastic sponge, in the tray at the upper right

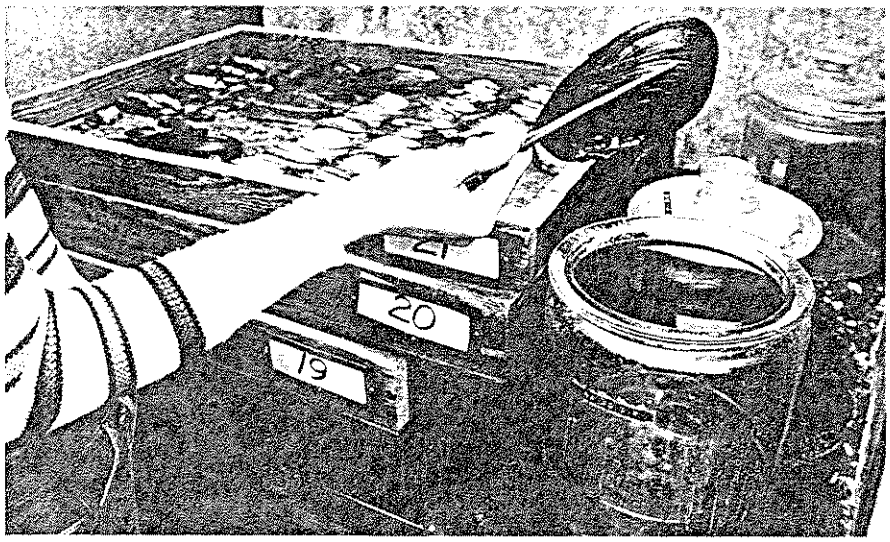


Figure 11. After they are numbered, bivalve shells are dipped in a xylene-paraffin solution and allowed to dry. This leaves a thin film of paraffin on the shell surface, which retards flaking of the periostracum and cracking of the shell

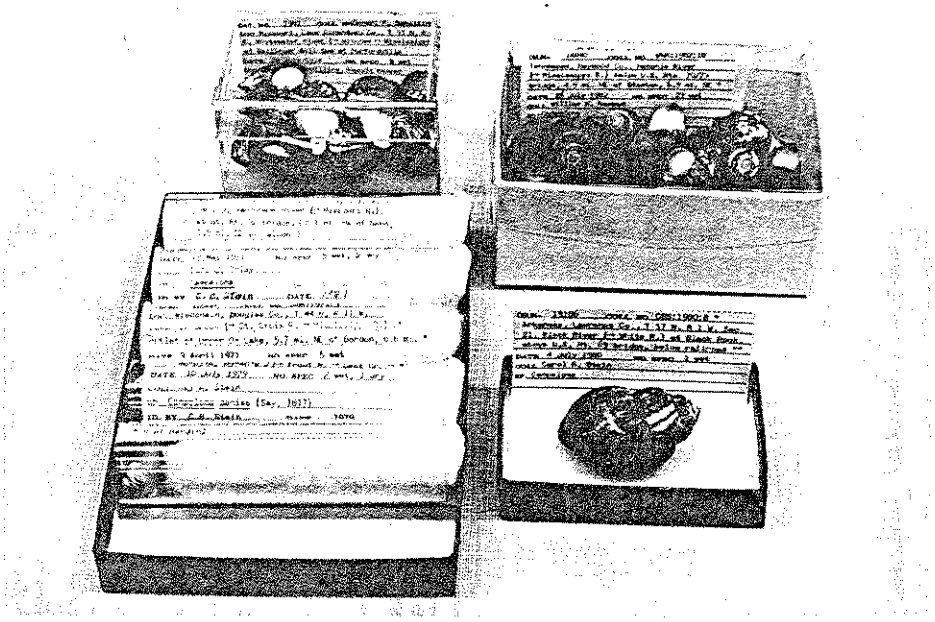


Figure 12. Fully-processed gastropod shells are stored with their labels in four different types of containers. Clockwise, from lower left, these are: 1. glass eight-dram shell vials, six vials per tray (when there are many vials of one taxon, we place stand-up cards at the back of the tray to indicate the river system represented in that tray); 2. hinged-lid plastic box with standup label and plastic sponge cushion; 3. pasteboard box with lift-off acetate lid, standup label, and plastic sponge cushion; 4. open pasteboard tray with standup label and plastic sponge cushion, used for large shells only

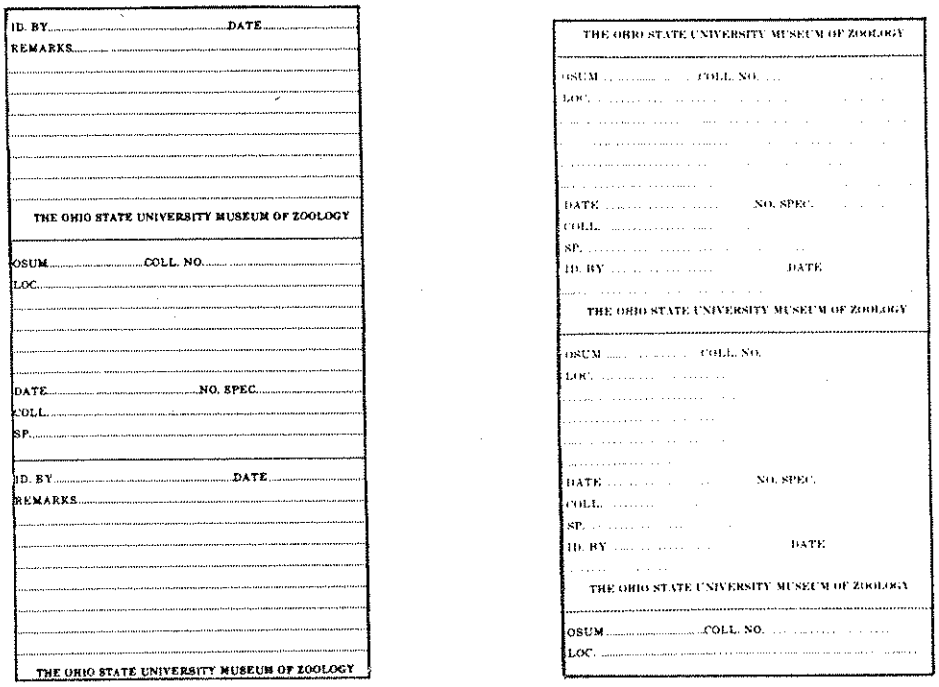


Figure 13. The two types of permanent lot labels used in the Museum's mollusk collections, printed in strip form for ease of typing: at left are standup labels printed on long-lasting card stock; at the right are labels printed on rag-content bond paper used for all wet specimens and all dry gastropod and sphaeriid lots kept in shell vials. After they are typed, the labels are cut apart on the dotted line immediately beneath the Museum's name. The standup labels are folded on the solid line in the middle of each label

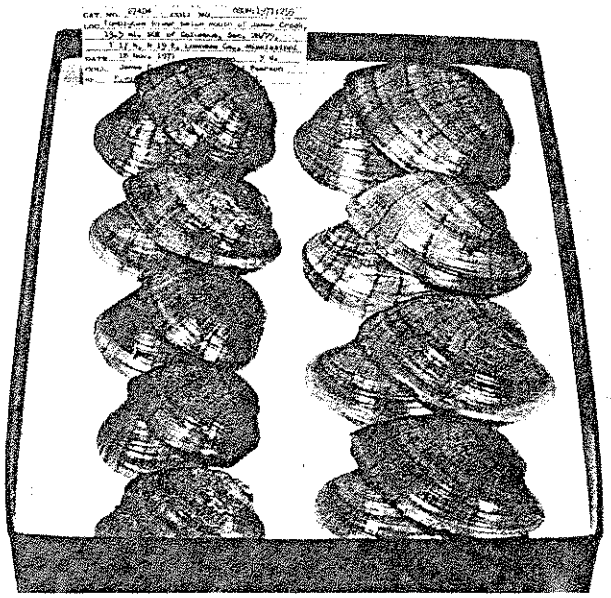


Figure 14. The consistent position and orientation of the shells in this cabinet-ready lot make it very easy to compare most of the features of the specimens without even touching them; also, the shells rarely move from this stable position when the tray is jostled accidentally

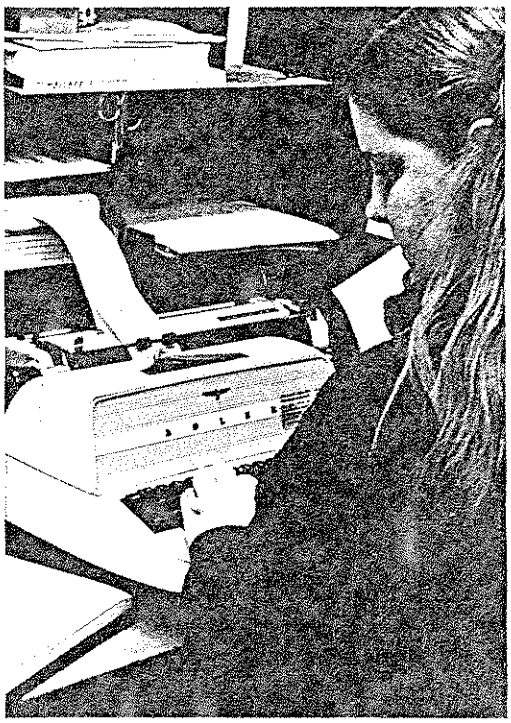


Figure 15. Using a special label micro-typewriter which prints 17 characters/in. and uses a carbon ribbon, Kathy G. Borror types a strip of specimen lot labels from data entered in the catalog book

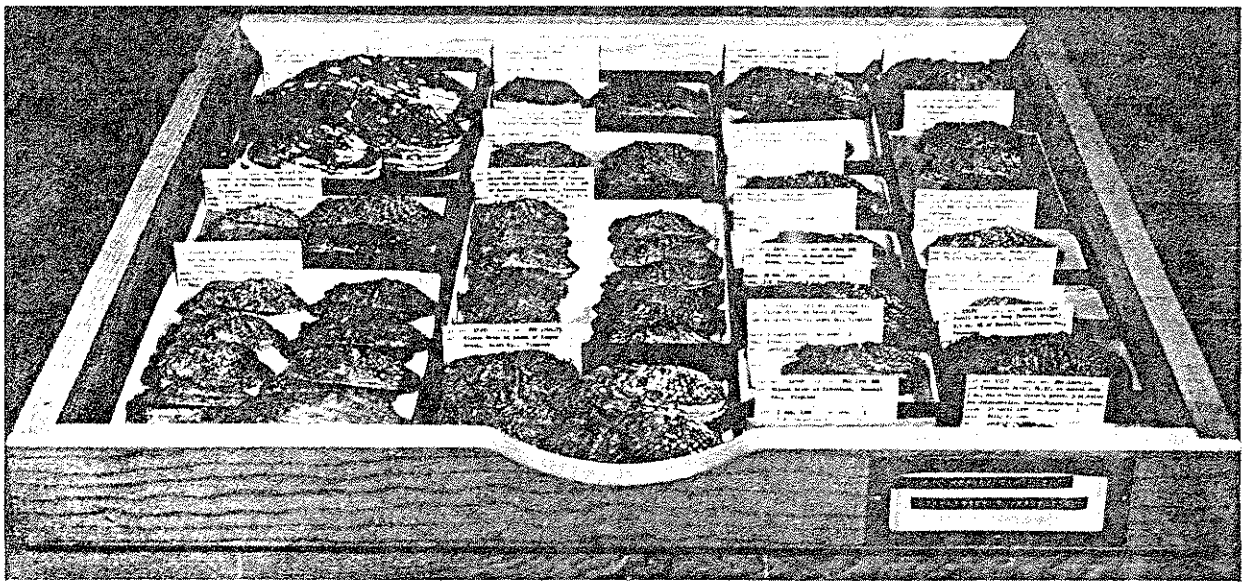


Figure 16. A quick scan of the rows of standup labels in this typical drawer of unionids reveals the shell lots by catalog number, the localities from which the various lots were collected, the dates of collection, names of collectors, and number of wet and dry specimens in each lot. The label on the front of the drawer identifies the species and the drainage system the specimens are from

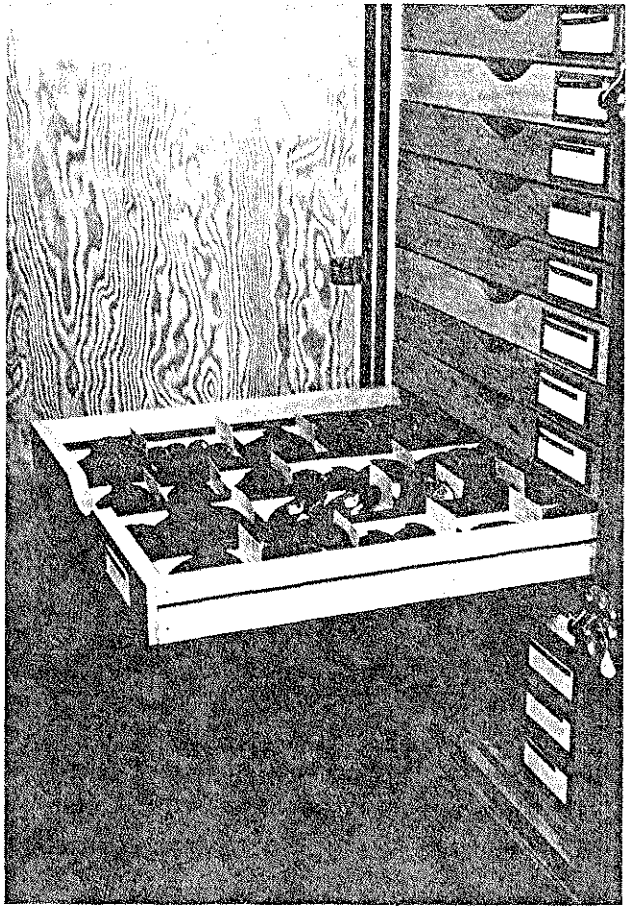


Figure 17. Modular trays fit neatly into the wooden drawers which are interchangeable throughout the Museum. The channels in the sides of the drawers fit over wooden rails in the cabinets which hold the drawers securely even when they are pulled out more than halfway. The 2-in.-deep drawers are adequate for nearly all bivalves and non-marine gastropods. For very large shells, we simply leave out one or more drawers above the drawer in which they rest

Figure 18. This view down the central corridor of the OSUM Bivalve Research Collection range shows some of the standard wooden cabinets in the left foreground. Behind them are shelves of processed wet material. In the far distance is the entrance to the room of partially processed collections. On the right are the ends of cabinet rows, with part of the processing area in the right foreground

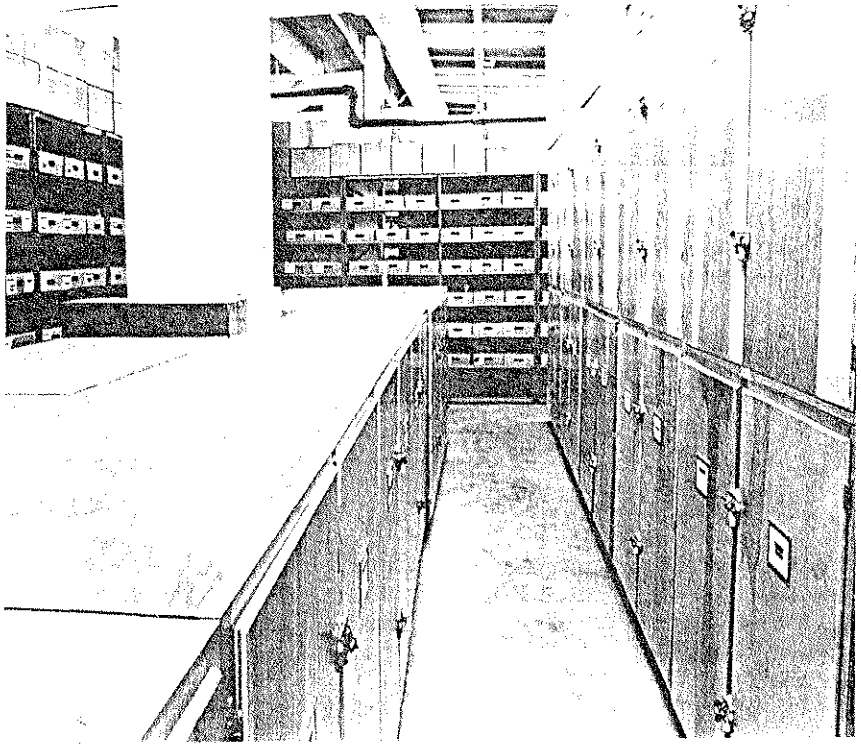


Figure 19. The OSUM Gastropod Research Collection is housed in standard wooden cabinets, with wet specimens in trays on the steel shelving in the background. It is adjacent to, but separate from, the Bivalve Research Collection

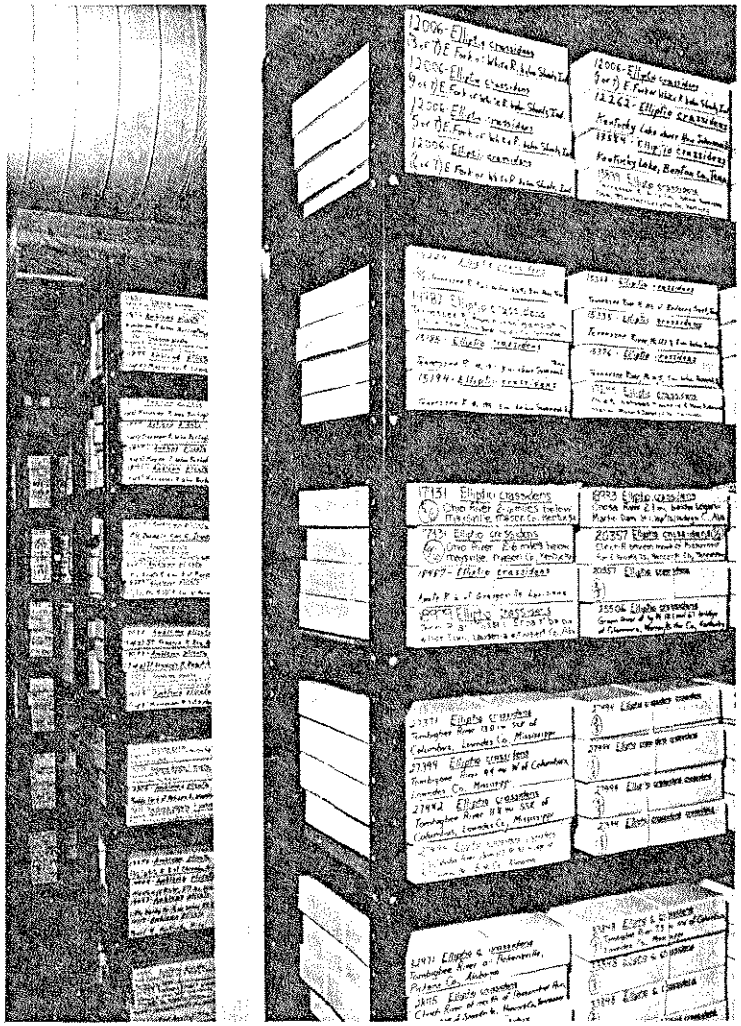


Figure 20. Large lots of unionids are housed in this very efficient and relatively low-cost storage system. The catalog number and species name are printed on the end of each box, and the first box of each lot also shows the locality where the shells were collected

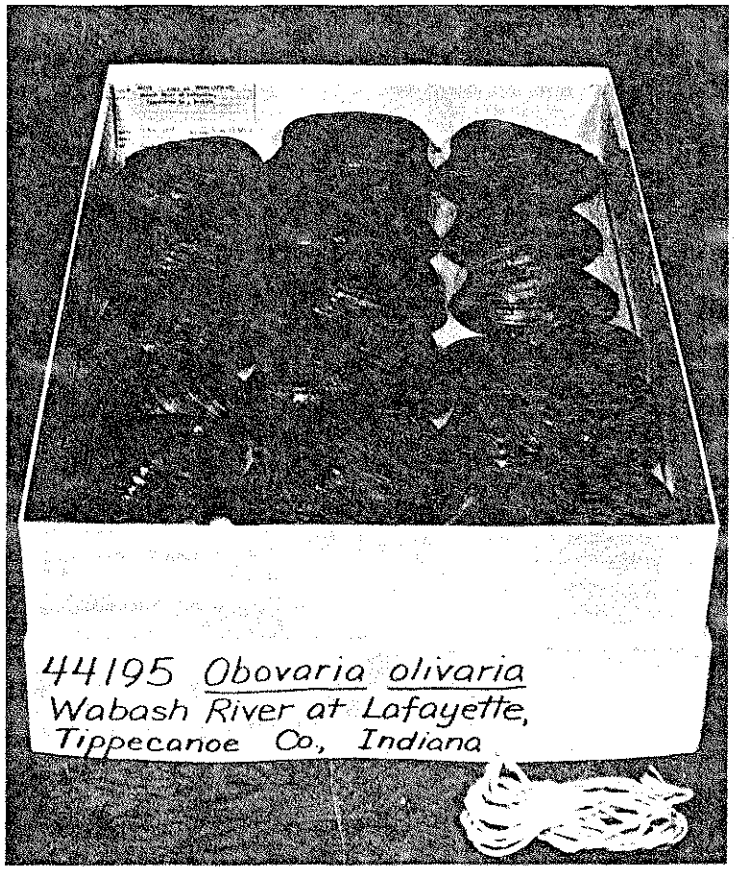


Figure 21. Large lots kept in closed boxes are packed in the same way as the ones in the cabinets, with a plastic sponge sheet on the bottom and a standup label at the back of the box. To keep the shells from shifting, and to cushion them from the weight of other boxes of shells, one or two full sheets of crumpled newspaper are carefully spread in the box lid before it is placed over the shells and tied

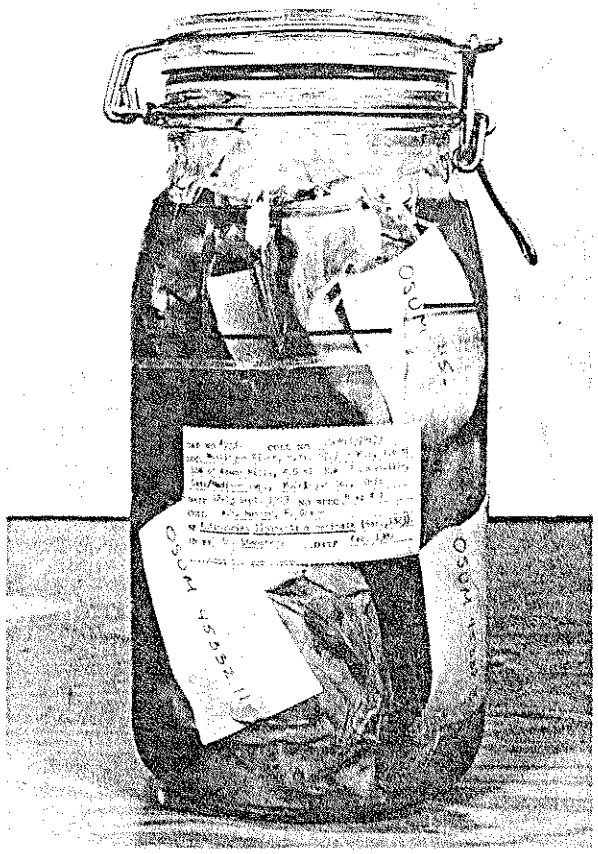


Figure 22. This fully processed lot of wet specimens is ready to be put into the collection. Each unionid has been carefully dissected from its shell, which is now in the cabinet or a box. The soft body, together with a label bearing the catalog number of that specimen (which is also written on each valve of the shell), has been placed inside an individual plastic envelope. All of the individually labeled bodies are placed inside the rubber-gasketed clamp-top glass jar, together with the typed OSUM label for that lot

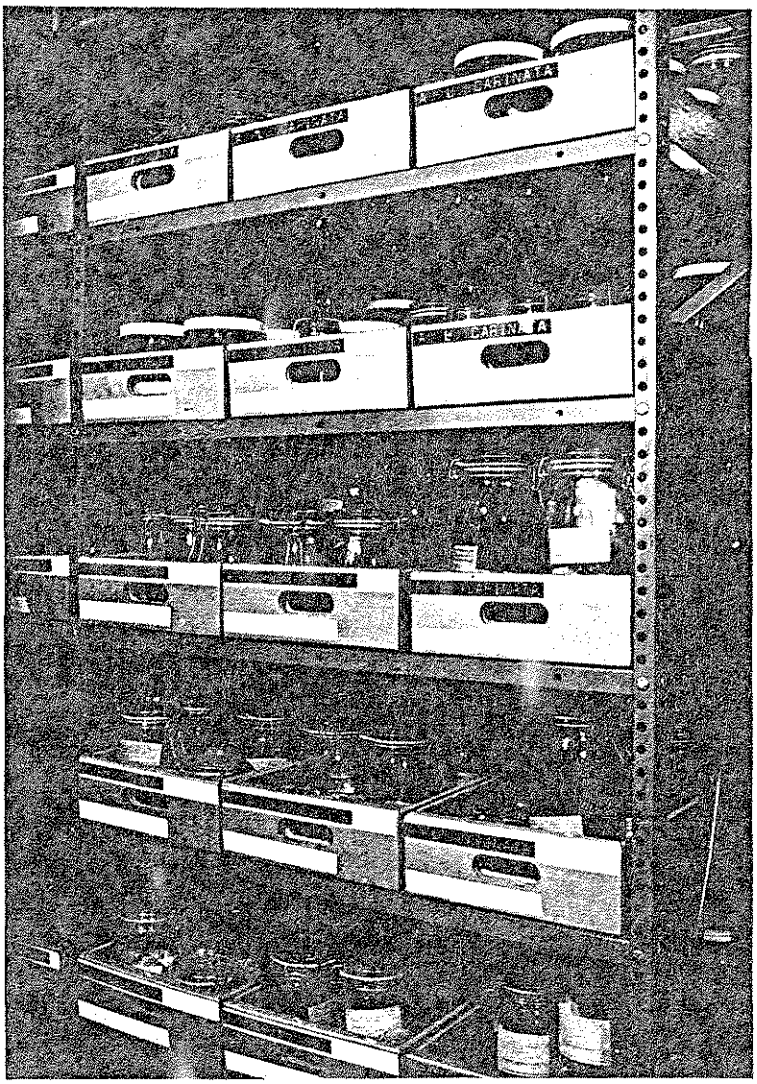


Figure 23. Cataloged wet specimens are kept in labeled trays on steel shelving. Eventually, locale labels will be added to the trays for convenience in locating the specimens

NOTES ON THE RELAXATION AND FIXING OF NAIAD MOLLUSKS FOR STUDY

by
Daniel Bereza
Academy of Natural Sciences of Philadelphia
Philadelphia, Pa.
Nov. 1972

(1) Place animals in receptacle and cover with water (just enough to cover them). Do not crowd; make sure each specimen can fully extend its foot and posterior mantle margin modifications without touching any other individuals.

(2) Put in a large pinch or two (0.3-0.5% solution) of Sodium pentobarbital (Nembutal Sodium) in receptacle for every gallon (or so) of water contained.

(3) Keep in this medium until bivalves no longer retract their foot and papillae upon stimulation with finger, pencil etc.

(4) Gently lift naiades by holding centers of valves between thumb and forefinger, anterior side down so that the foot hangs freely. (Do not press valves together).

(5) Place specimen in 5-7.5% formalin such that the protruding foot does not touch the bottom or wall of the formalin receptacle. (If specimen is not completely anesthesized, touching the foot at this point may elicit a reflex which retracts the foot and closes the valves.)

(6) Keep specimen in this solution for about 12 hours.

(7) Take specimen out of formalin and rinse thoroughly in a bucket of clean water.

(8) Place specimen in 30% ethanol and in time intervals of 1 hour, keep adding 95% ethanol in several portions, such that the preserving solution of the specimens becomes 70-80% ethanol.

Rules to remember

(1) The larger the animal, the longer it will take to anesthesize it.

(2) Certain Elliptio, Anodonta and amblemine genera are more resistant to the effects of Nembutal. Add more Nembutal in small pinches in several hour intervals.

(3) Also, amblemine genera have a narrow gape, therefore check for anesthesia even in cases of tiny slits as seen often in Cyclonaias.

(4) Add more Nembutal in acidic waters, as this compound deteriorates rapidly in such water.

(5) When testing for depth of anesthesia, do not look for complete loss of irritability to touch, because at this stage the animal is dead.

A GUIDE TO CHECKING LOCALE LABELS OF INCOMING COLLECTIONS TO THE OHIO STATE UNIVERSITY MUSEUM OF ZOOLOGY

For use by the staff and students of the Division of Bivalve Mollusks

A standard locale label used by the Bivalve Division of OSUMZ includes the following items of information, where known and applicable, in the following order:

1. Body of water
2. Bridge or mouth of another river
3. Distance and direction from nearest town of any size
4. Distance and direction from nearest LARGE town or city
   (preferably but not necessarily in the same county as the collection site)
5. Section (quarter section if possible)
6. Track and range
7. Township
8. County
9. Country (if other than U.S.A.)

state

10. Date of collection
11. Collector(s) (full names)
12. The collector's original field number
13. The drainage of the body of water collected

The only abbreviation permitted are:

miles = mi.
Township = Twp.
County = Co.
Section = Sec.
Track = T
Range = R
months = Jan. April July Oct.
Feb. May Aug. Nov.
March June Sept. Dec.
directions = N NNE NE ENE E ESE SE SSE S SSW SW WSW W WNW NW NNW
states = can be abbreviated ONLY as part of the name of a state route, such as Oh.Rt. 21, or Ky.Rt. 31W

Dates are always written in the form 1 Sept. 1949

NOT: Sept. 1, 1949 or 1/9/49 or 9/1/49 or IX:9:1949 ; these lead to confusion.

Distances are measured to the nearest tenth of a mile, from the collection site to the center of the town. We define the center of a town as the downtown area, or the largest major intersection. For instance, the center of Columbus would be the intersection of Broad and High Streets. Most, but not all county road maps show a dot at the major intersection of a town. Distances are never measured from the corporation or city limits, as this limit changes as the city grows. If a collection is made at a very small town the locale label would read "Larkin Fork Paint Rock River at Francisco,....." If the collection site is within the limits of a larger town or city, a typical label would read, "North Fork Licking River at S edge of Utica,....." The third case is that of a collection site within the city limits of a very large city. One can see that "Olentangy River at Columbus" could be anywhere along an 11 mile stretch of river. So for greater precision, our label would read, "Olentangy River at Oh.Rt. 161 bridge, 9.1 mi. N of center of Columbus,....."

SPECIAL CASES:

If the collection site is in or along an impounded stream, the label would read, for example
"Tennessee River at Kentucky Lake impoundment,....etc."
or "Olentangy River at Delaware Reservoir,....etc."
If the collection site is on the shore of a lake, the label might read
Lake Waccamaw, N shore,....etc."

Different maps vary greatly in accuracy, and the same type of map from different parts of the country will vary also. The most consistently accurate set of maps in the OSUMZ map library are the USGS 7.5 minute topographic maps. So whenever there is any discrepancy as to distances, locations of streams or names of streams, make out the locale label according to the USGS 7.5' map. If the 7.5' map for the collection site has not been published, or is not yet in the OSUMZ library, the second best "court of last resort" is our set of USGS National Topographic Maps. County road maps are very useful but not as consistently reliable as USGS maps.

NOTE: Any information ADDED to or CHANGED from the information on the original label is bracketed [....] on the OSUMZ Bivalve Div. standard locale label.

K.G. Borror, June 1977

INSTRUCTIONS FOR PUTTING AWAY NAIAD SHELLS IN THE OHIO STATE MUSEUM

by Carol B. Stein, September 1968

I. BOXES

A. Use the smallest size of box in which the shells will fit when arranged as explained below. Space is always at a premium in an active museum!

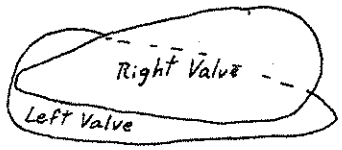
B. If the lot of shells will not fit properly into one of the four sizes of boxes (frequently referred to as trays) which are used in the cabinets, then use a white shirt box. See the special instructions for putting away naiades in shirt boxes.

C. Always place a piece of plastic foam of appropriate size in the bottom of the box before adding the shells or the label. Exception: When the smallest size of box is used, the bottom half of the folded label is placed on the bottom of the box and the foam is placed on top of it. In all other boxes the label is placed on top of the foam at the left rear corner of the box.

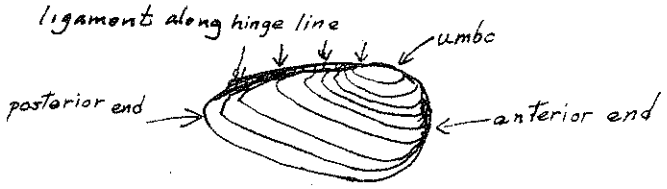
II. SHELL ARRANGEMENT

A. A shell may be placed in a box in one of the following two positions:

1. (This position is preferable, if space allows, but if you can use a smaller box by using the other position, then do so). Place the left valve flat against the foam, nacre down, with the hinge line approximately parallel to the top of the box. The right valve is then placed over the left valve, with its anterior end and hinge line resting on the foam. (HINT: the umbones point toward the anterior end, which is usually the more rounded end of the shell)

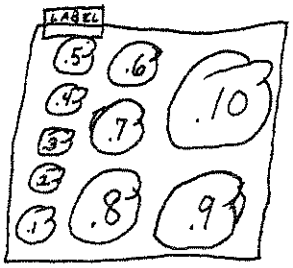


2. Fit both valves together in their natural position. Place the shell on the foam with the hinge line parallel to the top of the box and the anterior end toward the right-hand side of the box. (NOTE: in this position, as in position 1, the right valve is always uppermost and is oriented in the same way. This makes for neat, aesthetically-pleasing drawers. But, more important, it saves a great amount of time later on for anyone who is studying the species because he can quickly compare large numbers of shells at a glance without having to take them out of the boxes and match up the valves from a helter-skelter arrangement).



3. Compromise: If there are too many shells to use Position 1 entirely, but too few to fill the box neatly with Position 2, then use Position 1 for the large specimens and Position 2 for the little ones.

B. The smallest specimen (numbered _____.1) is always placed at the lower left-hand corner of the box. The remaining shells are then placed in numerical sequence in vertical rows, starting at the lower left and running up to the top (back) of the box, then from the top to the bottom, snake-wise.



C. The number of rows is determined by the size of the specimens. Obviously, large specimens will not go into as many rows as small ones will.

D. If the shells are placed in Position 2 (valves together in their natural position), the ventral margin of the upper shell should be neatly tucked underneath the dorsal (hinge) edge of the preceding one. This helps to save space, and it also keeps the specimens more snugly in position in the box.

E. Likewise, if Position 2 (valves in their natural position) is used, the posterior ends of the specimens in the second row should be nestled in between the anterior ends of the specimens in the preceding row.

F. If Position 1 (one valve overlapping the other) is used, then each specimen should rest directly on the foam. It should not overlap or touch another specimen.

III. PLACEMENT IN CABINETS

A. The species are arranged in phylogenetic sequence within the cabinets, corresponding to the posted lists. The cabinets begin in the naiad range extension and are continued in the naiad range. In each row of cabinets the drawers are arranged from the top drawer to the bottom drawer in each stack, and the stacks are arranged from left to right, just as in a library. Each cabinet door bears a label listing the genera and species contained within that unit.

B. Every drawer is marked with a species name, author, and date. Be <u>sure</u> to check the label of your specimens against this. If it does not match, don't put it in---find out what is wrong. Remember, a set of specimens in the wrong drawer may remain "lost" for years, and it could cause a lot of trouble if a researcher couldn't find it when it was needed.

C. All drawers containing specimens of a given species should be completely filled except the one or two on the bottom. New specimens are added to the unfilled drawers. If you have to start a new drawer, be sure that it has a label. Sometimes there will be extra labels in the last drawer containing boxes of that species. If not, then make a new one. Be sure it is the same as the others.

D. Arrangement of boxes in drawers:

1. Boxes are arranged in vertical rows, back to front.

2. All boxes in a given row are of the same width, but may vary in length from front to back.

3. Rows should be so arranged as to make maximum use of the width of the drawer.

IV. SHIRT-BOXED SETS OF NAIADES

A. Packing the box

1. Use as few boxes as possible, but arrange the shells as explained above.

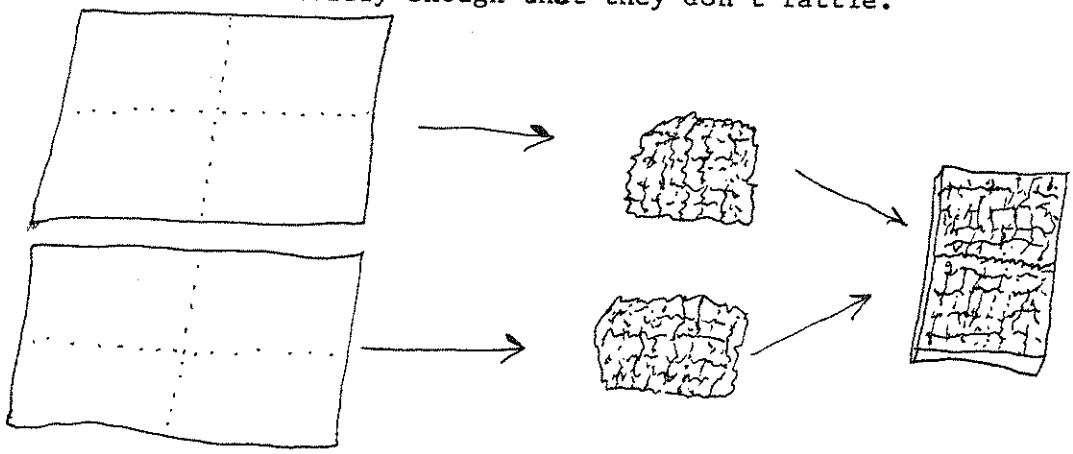
2. Place an appropriate-sized piece of plastic foam in the bottom of the box.

3. Place the folded stand-up label in the left rear corner of the first box. The other boxes do not have labels inside them. If there is an original or field label, place it on top of the base of the stand-up label.

4. Arrange shells as explained above. If the set is too large to fit in one box, continue it in the next one. The smallest shells and the label are always in the first box, and the largest shells are always in the last box.

5. If there are very many small shells which might slip around in the box, place a sheet of plastic foam over the shells before cushioning the box lid with newspapers. Large shells will be held gently but firmly in place with properly crumpled newspapers; they do not need a layer of foam over them. (this saves $$$!.....We need $$$$ for other things --- like wages....)

6. Cushioning: Crumple at least two, and preferably three, full-size sheets of newspaper both vertically and horizontally. Then adjust these crumpled paper cushions into the box lid so that they completely fill the lid. Please do not use anything smaller than a full sheet of newspaper in these boxes. Magazine sections are fine for wrapping packets of shells in the field, but they are hard to manage when opening and closing these shirt boxes in research work. Use as many full-size sheets as necessary to pack the shells securely enough that they don't rattle.



7. Labeling the ends of the boxes:

   a. Place the catalog number in the upper left-hand corner of the end of each box in the set, using a fine-tipped black felt marking pen.

   b. Place the scientific name of the specimens in the upper right-hand corner, but omit the author and date. Otherwise, the scientific name is written just as it appears on the label inside the first box of the set. If the word form is used on the label, it may be abbreviated f., and it is not to be underlined. All of the rest of the name is underlined, however. Remember that the name of the genus is capitalized, but the species and the form or subspecies name must not be capitalized. The scientific name should be placed on each box of the set.

c. If more than one box is used for the set, then a circle is drawn beneath the catalog number at the lower left corner of each box. Inside this circle a fraction is written. The lower number (denominator of the fraction is the total number of boxes in which the set is packed. The upper number (numerator) is the number of this particular box in the set. For example, if there are 3 boxes in a set, the one with the smallest specimens (and the label) is numbered (1/3) ; the middle one is numbered (2/3) ; and the last box is (3/3) .

d. In the remaining space on the end of the first box you will write the locality where the specimens were collected. Look at the locality typed on the label in the first box and at the space available and decide whether you can get all of the locality data neatly and legibly printed in the space available, using no more than two lines of printing. If so, fine!

e. If there is too much information on the label for the space available on the box, then be sure that the following information is printed on the end of the box:

1. Name of the body of water. Please do not abbreviate any parts of the name. Exceptions: R. may be used as an abbreviation for River
Cr. " " " " " " " Creek

2. Distance and direction from nearest town. The standard symbols N, SW, ENE, etc. may be used for directions, and mi. may be used for mile or miles.

3. County. Write out the full name of the county. Co. may be used for County and Par. may be used for Parish.

4. State or province. Long ones may be abbreviated if necessary (see standard list of abbreviations and symbols), but please write out the short ones like Ohio and Iowa.

f. Locality information is given only on the first box of each set.

g. Example of properly labeled set of two boxes:

40698 *Amblema plicata f. costata* (1/2) *Grand River 3.2 mi. NE of Berne, Blue Twp., Lincoln Co., Arkansas*	40698 *Amblema plicata f. costata* (2/2)

8. Tying shirt boxes:

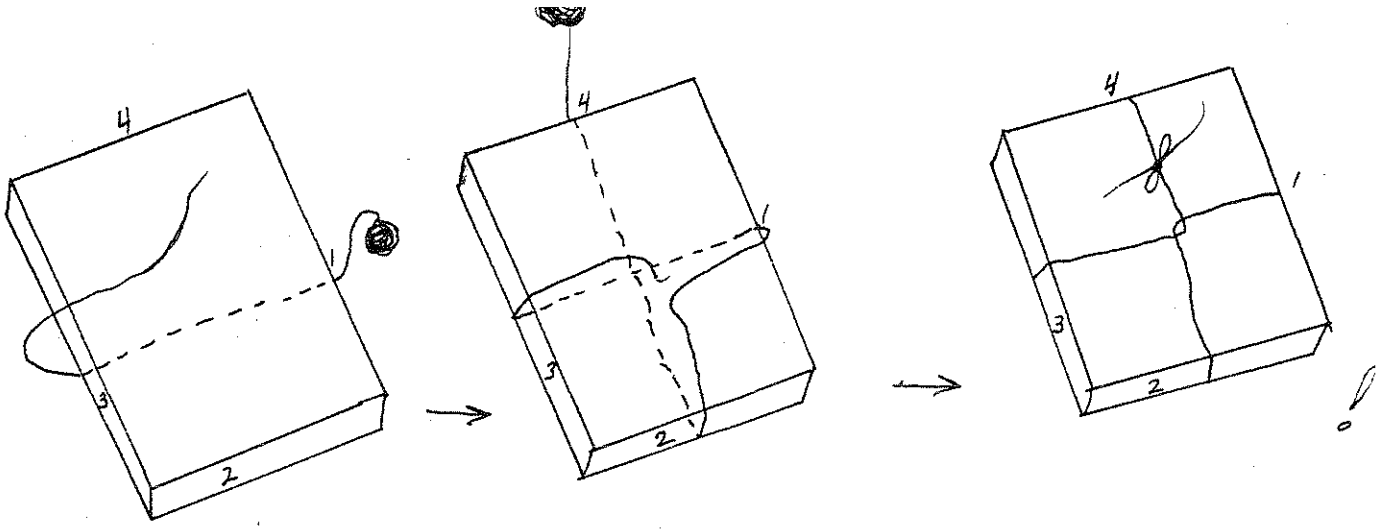
a. Tie each box individually with 24-ply unpolished cotton twine.

b. Method of tying:

1. Pull free end of string beneath box from side 1 to side 3.

2. Hold box up by the string while passing the long end beneath the box from end 2 to end 4.

3. Pass the short end of string (from side 3) through the loop 1 - 2 and tie it in a shoelace-style bow to the long end coming up from end 4. Do not tie the string in a knot. Please leave plenty of string in the bow so it can easily be re-tied when the box is opened.



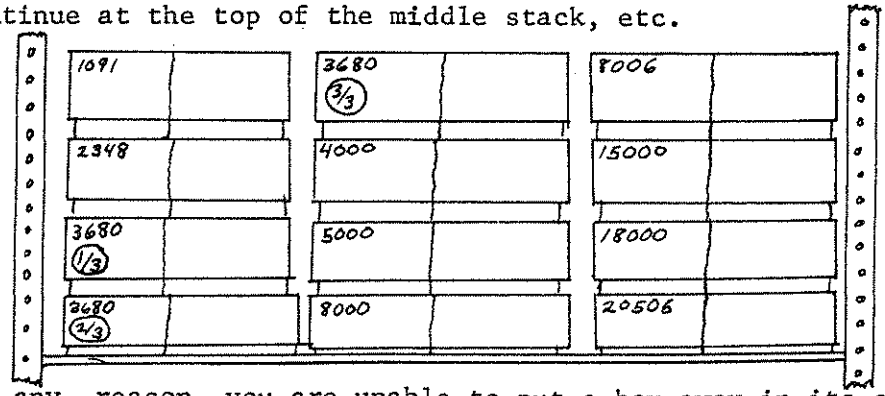
8. Putting shirt boxed sets away:

   a. As in the rest of the collection (except for the 3-gallon jars), the shirt-boxed specimens are arranged phylogenetically by genus and species. Check the diagram posted in the naiad range extension to see the sequence.

   b. Each vertical row of shelves is one shelf unit. The shelf units are arranged beginning at the far left in the front of the naiad range extension and continue in phylogenetic sequence from left to right around each group of shelves all the way around the room.

   c. Within each shelf unit, the shelves are arranged in sequence from top to bottom, then to the top of the next shelf unit to the right.

   d. Each species (and often each subspecies or form --- check the shelves carefully to see) is arranged on the shelves in strictly numerical sequence. The numbers begin at the top of the left-hand stack of boxes on a shelf, go down to the bottom box in that stack, continue at the top of the middle stack, etc.



9. If, for any reason, you are unable to put a box away in its correct position or are in doubt about where it should go, please do not leave it in the naiad range extension. It might get lost for years! Bring it back to the cataloging bench until it can be put away properly.

10. Whenever you put a box away, please check the last several boxes of that species to be sure they have not gotten out of order. It is extremely important that the sets of specimens can be found quickly and easily.

11. All Lake Erie specimens are to be put away in Dr. Stansbery's office.

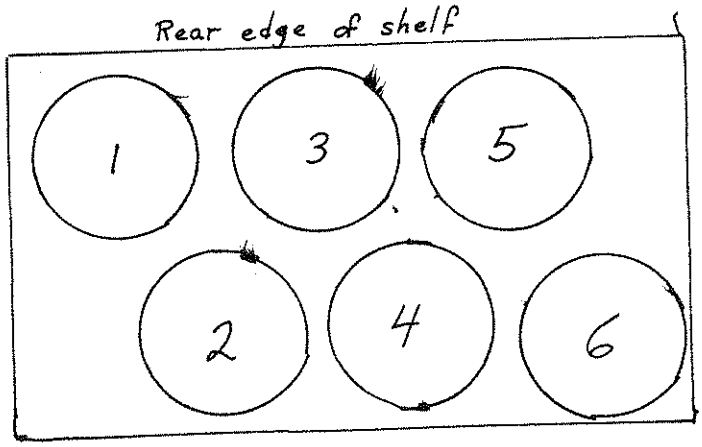
INSTRUCTIONS FOR PUTTING AWAY THREE-GALLON GLASS JARS OF NAIADES

1. In addition to the regular typed and metal labels inside the jar, a 3 x 5" card is cut in half crosswise and the catalog number, jar number, and scientific name are printed with a felt marker. This label is fastened with a loop of masking tape to the neck of the jar about an inch or so below the edge of the lid.

2. The jars are placed in numerical order on the bottom shelves of the green shelf units in the naiad range extension. For sequence of shelves to be used, see the chart posted in the naiad range extension.

3. Place the jar so that the outside label can most easily be read from the aisle in front of the shelf.

4. Place six jars on each shelf, staggered as shown below to allow maximum space between jars. These jars are <u>very</u> fragile and often break if they just gently nudge each other! Please use great care in handling them so that you will not be injured by broken glass.



20918 (1/2)
Actinonaias
carinata

sample outer label

C.B. Stein, Sept. 1968

TECHNIQUE FOR PRESERVING THE PERIOSTRACUM OF NAIAD MOLLUSKS IN COLLECTIONS

Kathy Gail Borror
Museum of Zoology
The Ohio State University
Columbus, Ohio 43210
February 1977
Revised December 1982

1) In a large-mouth glass jar, place 1 gallon xylene and 1/4 lb paraffin (regular canning wax).

2) Let stand 36-48 hours, or until paraffin is dissolved; stir.

3) In a well-ventilated area (preferably under a hood), use an 8" - 10" pair of forceps to dip naiad specimens into mixture, shaking several times to remove all excess liquid.

4) Lay the dipped specimens nacre-down in a tray made of wood, masonite, or some other porous material. If a non-porous surface is used, cover with paper toweling.

5) Let dry in an area with good air circulation.

Specimens in the bivalve research collection at OSUM have been processed in this way to help prevent the periostracum of specimens from flaking off and, to some extent, the shells from cracking. Painting the posterior portion of the nacre with a clear-drying, water soluble glue such as Franklin "Titebond" does much to strengthen very thin or fragile shells, such as Cumberlandia monodonta (Say, 1829) or the posterior expansion of the females of some species of the Genus Epioblasma. Glue is not applied to the periostracum since this would alter its texture - a character of value in systematic studies.

We have found this method to be superior to several others we have tried or encountered. Most importantly, the thin layer of paraffin on the shell does not alter its natural appearance. Shellac, varnish and plastic coatings not only may yellow with age, but give treated specimens a uniform gloss, completely obliterating the natural, variable texture of the periostracum.

Other techniques sometimes used include rubbing the shell with oil or petroleum jelly. Oil, however, tends to slightly darken the periostracum after several years, and both of these coatings can easily stain specimen labels and trays, and collect dust and dirt. Dipping specimens in the above described xylene-paraffin mixture is much less time consuming and seems to have none of these disadvantages.

This method of protecting the periostracum of shells was first described and used by Dr. William J. Clench (1931:30) at the Museum of Comparative Zoology at Harvard University. It was later cited by Dr. Henry van der Schalie (1941:13; 1961:65) in the American Malacological Union Symposium on "How to Collect Shells."

LITERATURE CITED

Clench, W. J.
1931. A preventive for the scaling of the periostracum. Nautilus 45(1):30-31.

van der Schalie, H.
1941. On collecting fresh water mussels. Ann. Rept. Amer. Malacol. Union for 1941:9-14.

van der Schalie, H.
1961. Fresh water mussels, pp. 60-66 IN How to collect shells: A symposium. Amer. Malacol. Publication, 92 pp.

Ohio State University Museum of Zoology Report 1980 (77) 1-12

A SHELL CLEANING MANUAL

by
Jon E. Ditmars
August 1980

This manual describes the materials and techniques found to be helpful in removing various environmental deposits on bivalve mollusks for the research collection at The Ohio State University Museum of Zoology over the past decade.

General Equipment

Sink - This is stainless steel and has a special drain trap which unscrews to empty silt and debris that accumulates regularly. Sponge out sink daily to prevent buildup of limy deposits. If some deposits do accumulate, remove with Lime-A-Way.

Faucet - The faucet was extended by attaching another piece of pipe which can be moved about like the arm of a dentist's drill. At the end is a standard lab spout which makes rinsing easier by directing a steady stream of water onto the specimen being cleaned. If desired, a rubber mat or some device to break the fall of the water (such as an oil-based clay slide) is useful in muffling the sound of the water against the sink.

Misc. - Standard lab pegboard for hanging equipment and drying same, sponge plastic pad to keep brushes and tools drained while using them, unbreakable container for powder shell cleanser, cloth towels and a sponge, Johnson's Baby Powder, surgical and general purpose rubber gloves, dust nose mask, magnifying lamp.

Cleaning Tools

Nylon tooth brushes - We use three or four row bristle brushes, both hard and soft. The soft brushes are used for cleaning under the umbo, in crevices and in the muscle scars, because the smaller, more flexible bristles

can reach into these areas more easily. Also, use soft brushes for cleaning delicate and subfossil bivalves. An art brush is helpful for reaching dirt in shells with very deep umbonal cavities. Hard brushes are used for general cleaning. When worn down, hard brushes work well for scrubbing tough dirt from shells.

Nylon hand brushes - These are used for removing loose dirt and clay from specimens with soft parts before preserving them (known as "rough cleaning")

Dental picks - There are a variety of picks and scrapers used to probe cracks and crevices in a shell, to remove stubborn deposits, and to loosen dried flesh from muscle scars.

Misc. - Other less frequently used tools include old credit cards cut in half and tooth brush butts. These flake off thick dirt deposits from shells and reduce cleaning time. Tissue probes work well with dental picks. A Rust-o-Razor and fiber glass brush may be helpful in special cleaning jobs, particularly in removing stains from nacre, but the Moto-Tool often does a better job than these.

Moto-Tool - See special instructions fro this.

General Techniques

Step One - Put on rubber or surgical gloves. Adjust faucet and container of cleanser within easy reach. Turn on water to a lukewarm and gentle to moderate stream. Wet brush, shake out excess water (important), and dip it into the cleanser so just a thin layer of powder adheres to the bristles. Any more is a waste of cleanser.

Step Two - This stage is called "rough cleaning" and is done to prepare wet specimens (= specimens having soft parts) for identification and preservation, or as a first step in "regular cleaning."

Wet specimens - No powdered cleanser is needed (or should be used) for

rough cleaning wet specimens. Quickly scrub off loose environment with intermittent rinsings until enough of the periostracum appears so the specimen can be easily identified, and also so no dirt comes off in the preservative.

Dry specimens - Use powdered cleanser in rough cleaning dry specimens. This procedure is used for large lots that will have to be culled, so that cleaning effort is not wasted. After the lot has been culled, it will be returned to the cleaner for regular cleaning. Rough cleaning should take less than a minute for smaller bivalves, about one minute for palm sized shells, and perhaps longer for larger naiades. Again, rough cleaning is simply removing all mud, clay, etc., but ignoring tough deposits.

Step Three - This is regular cleaning, and it is performed on all shells that will be added to the catalogued collections. Regular cleaning means scrubbing all the environment off the specimen. Wet the hard bristled toothbrush, shake out excess water, lightly dip into the cleanser, and brush the shell with the bristles fairly perpendicular to the surface. Make sure any fragile areas of the shell are supported by your hand from underneath. A gentle to moderate pressure and a rapid back-and-forth motion of the brush makes the best use of the powder's abrasive ability. Rinse after scrubbing a little to check how much environment has been removed. One indication that all the dirt has been removed is that the cleanser suds remain white. Also, stop cleaning when a deposit fails to yield to further regular cleaning, or if the periostracum (on the convex surface of the shell) begins to turn a lighter color. When the periostracum is scrubbed away so that it

becomes a lighter color than the natural color for that species, this is called "over cleaning." This is easy to do and the cleaner should be careful.

Some shells require little or no regular cleaning, but the shell preparator should skip step two and go to step three, because these shells are covered mostly with a tough deposit that only the moto-tool or other picks or scrapers can remove.

The soft toothbrush is used similarly as the hard brush described above, but mostly to clean the nacre. The hard brush can be used for the nacre too, particularly to remove tough dirt and stains. But the soft brush cleans the umbonal cavity, muscle scars, and in between the teeth better than the hard brush. The soft brush is best for cleaning all the surfaces of delicate shells or subfossil material; although the hard brush is more effective on such shells that are covered with stubborn dirt, and if scrubbed gently with the cleanser doing most of the work.

Regular cleaning may take several minutes for small shells, three to five minutes for a palm-sized one, and up to 20 minutes for the largest ones. Twenty minutes is about the maximum time limit to be spent on a shell unless told otherwise.

Step Four - This step is only necessary if small areas or specks of environment remain after finishing steps one through three. Some specimens become overcleaned easily, and this step probably should be skipped in such cases. Also, one should avoid spending a lot of time with "touching up" a shell, which means using the Moto-Tool, any of the picks or scrapers or other miscellaneous tools to remove stubborn deposits or stains. Per-

fection in cleaning is the ideal, but practically speaking, the realistic goal is to clean enough of the environment from a bivalve in order to correctly identify it, while preserving as much of the natural character of the specimen as possible. "Touch-up" shouldn't take but a few minutes at most unless the cleaner is told otherwise.

Special Cases

Algae - Algae is usually associated with marl deposits on a shell, but either one can be found separately. Sometimes a light growth of algae can be scrubbed off with either the tooth or hand brush and cleanser. However, if the algae rootlets have penetrated the periostracum, it will be difficult, or not worth the trouble, to remove all the algae. Large chunks of the algae are easily cut off with scissors or a scalpel. Lesser amounts can be scraped off with dental scrapers or rubbed off with the Moto-Tool. Stop before the periostracum begins to be over-cleaned. Algae stains the nacre green and this color cannot be removed. Cleaning time may be 5-10 minutes for a palm size naiad and 15-20 minutes for larger bivalves.

Marl - Marl in thick deposits will usually flake off rather easily. Use a dental scraper or tooth brush butt to do this. After this, remove most or all of the marl with a hand brush and/or toothbrush and cleanser. Ifany small specks of marl remain, remove them with a dental pick. Sometimes the Moto-Tool works well on marl. Avoid over cleaning or scratching the periostracum with these tools, however. A dilute solution (10%) of HCl will dissolve marl, but should be used only in special casses. Care must be taken because the HCl will dissolve the nacre of the shell as well as the marl, and will also bleach the periostracum. When using HCl, touch it to the marl deposit sparingly with cotton and forceps. Cleaning time for a marl-encrusted shell is about the same as

for a shell with algae on it.

Iron oxide - This deposit is reddish in color. Sometimes it is thick and soft and can be flaked off with a dental scraper or toothbrush butt. Once the bulk of the deposit is flaked off, brushing with the hard toothbrush and cleanser will generally remove all of the iron oxide deposit. If needed, use the Moto-Tool, dental tools, or Rust-o-Razor to touch up the shell. The iron oxide clay may be cemented to the shell surface, however, and after the initial rough cleaning, immediate use of the Moto-Tool and/or dental tools may be required. If the iron oxide clay is particularly stubborn, cleaning time could take 5-10 minutes for small shells, up to 20 minutes for palm-size bivalves, and 20-25 minutes for larger specimens. If it would take longer to clean thoroughly, check with the supervisor to determine the value of spending more time on such a shell.

Black deposits - Black or brown deposits may be loosely encrusted on the shell surface or they may be stubbornly cemented to it. For the former situation, most if not all the deposit can be removed by hand and tooth brushes and cleanser. A dental scraper, to flake off large chunks, may speed the cleaning process. For stubborn black deposits, after rough cleaning with a hand brush and cleanser, carefully brush the deposit away with the Moto-Tool and, if needed, dental scrapers. Stop before the shell surface begins to become over cleaned, or if more than 20 minutes is spent on palm size naiades or 25 minutes on larger shells.

Tar, Shellac, Oil, Bizarre stains - Since some shell specimens are collected from polluted bodies of water, a variety of deposits are found on them. Turpentine or a general use solvent will remove or loosen tar, shellac, and oil. Then use the hand and/or toothbrushes with powdered cleanser, and follow with dental tools or the Moto-Tool if needed. The shellac and

oil covered shells are from unpreferred preservative methods used by those who donated the specimens. Scrubbing with cleanser has proven effective on most other problem deposits or stains. However, some stains penetrate the shell and cannot be removed. Occasionally, a grayish metallic discoloration occurs, and should be ignored because it appears to be part of the naiad shell. Cleaning time shouldn't exceed 15-20 minutes for palm-sized specimens and 20-25 minutes for larger bivalves.

Estimated Cleaning Time

Time estimates are based on approximately 60 palm sized (3" diameter) shells per tray.

Light Cleaning - This is basically rough cleaning. There is usually little need for more than gentle brushing with the hand brush, soft tooth brush, and cleanser. Cleaning time: 1 - 2 1/2 hours per tray.

Moderate Cleaning - This is regular cleaning. This involves vigorous scrubbing with the hand and tooth brushes and cleanser. It may include some use of the dental tools or the Moto-Tool, but here these are mainly for touch up. Cleaning time: 2 1/2 - 4 hours per tray.

Difficult Cleaning - This is strenuous regular cleaning with the brushes and cleanser. However, much of the time will be spent using the dental scrapers and Moto-Tool. Cleaning time: 4-8 hours.

REMOVING THE ENVIRONMENT FROM BIVALVE MOLLUSKS

UPDATE 1980

by Jon E. Ditmars.

This update is made necessary by the addition of the Dremel Moto-Tool to bivalve cleaning tools. Hopefully, the shell preparator will become more adept at using the Moto-Tool after reading these instructions.

The shell preparator should first realize that it is possible to over-clean (scrub away part of the shell surface) even with a soft bristled tooth brush on some types of bivalves. So care should be taken especially with the Moto-Tool, which has metal brushes that can easily scar a shell surface because of much greater hardness of the metal versus the periostracum. The rapid rotation of the brush head also makes it difficult to gauge when one reaches the shell surface while cleaning away the dirt.

Generally, the Moto-Tool should be used ONLY for removing very stubborn dirt, carbonate or oxide deposits. A good rule to remember is: if the shell can be cleaned in a reasonable time without using the Moto-Tool, then don't use it.

Usually a good procedure for operating the Moto-Tool is as follows:

1) First, use the boot brush to rough clean the shell specimen.
2) Then use the tooth brushes to remove the rest of the environment from the shell.
3) If any stubborn dirt remains use either the dental tools or Moto-Tool, or both to finish the job. If only a small amount of dirt remains, say around the beak sculpture, generally a dental tool to loosen the deposit is adequate. If a larger amount remains, then using the Moto-Tool speeds the cleaning process considerably.

Therefore, the best technique for grinding away tough environment from a bivalve surface is a combination of a gentle sweeping motion back and forth over the hard dirt deposit while lightly touching the shell with the Moto-Tool.

Only a very slight pressure is needed to wear away the encrustation since the spinning wire bristles cut through the dirt much faster than one might expect. Also, after grinding a little of the deposit away, rinse away the loose debris and grind some more, and again rinse, alternating thusly until the bulk of the dirt is removed. Sometimes a dental pick and tooth brush are needed to touch up a shell after using the Moto-Tool to clean small specks of dirt in cracks or grooves on the shell surface.

Sometimes the color of the periostracum is lighter than the surrounding area due, it seems, to being buried by the deposit so long. However, other times the lighter color is a result of over cleaning, which is particularly evident with shells which have delicate periostracums. In such cases, it is best not to use the Moto-Tool.

The Moto-Tool is most effective in loosening dirt deposits that are too hard to brush away with cleanser, too stubborn to pick away rapidly with a dental tool, and rather thick. If the dirt deposit is razor thin and too difficult to pick away, it may be better to leave the specimen alone rather than overclean it. However, if one has a steady hand, it may be worth trying the Moto-Tool in such cases.

There are a few other tips in using the Moto-Tool the preparator should know. Change the metal brush head when it wears down between 1/8" and 1/16". There are two types of metal brush heads used regularly. The flat, circular one (No. 428) seems better suited for getting between ridges. The cone-like head (No. 442) is better at heavy duty jobs. The diamond tipped brush head was intended for rare cases where a heavily encrusted specimen resisted cleaning even with the Moto-Tool. Such a shell might be covered with tar or shellac, for instance. Of course, particular care should be taken when using this head since it could easily cut right through the shell.

Adjust the Dremel Moto-Tool's speed control dial to suit the lot of

shells being cleaned. For instance, it might be helpful to increase the speed to overcome the torque resistance due to a particularly stubborn kind of dirt deposit on the shells; or a slower speed might be better while cleaning fragile shells. One of the most delicate and frustrating assignments is removing a hard deposit from a fragile shell. Sometimes it is best not to finish the job rather than to break the specimen.

Oil the area around the drive shaft where the brush head is inserted if it looks rusty.

CAUTION: Don't adjust the Moto-Tool plugs with wet hands or while the faucet is on.

Brushes may be purchased at Columbus Hardware.

SOME METHODS AND MATERIALS USED IN THE PRESERVATION OF BIOLOGICAL SPECIMENS

The objective of the preservation of plant and animal forms is their maintenance for future reference and study. Such information as identification, measurements, counts, stomach analysis and anatomy can frequently be studied best with the use of preserved specimens. Curatorial techniques which best preserve the qualities listed above (or others sought) are consequently the most desireable. Most forms are best kept by immersion in (and occasionally by injection with) a preserving fluid while some forms (i.e. adult terrestrial insects, shells, bones, pelts, and plants) may be best stored in a dry state. The fluids described below are easily made and have given satisfactory results when used as indicated.

ARTHROPOD FIXATIVE AND PRESERVATIVE

Ethanol	75%	15 quarts	
Glycerine	5%	1 quart	= 5 gallons
Distilled Water	20%	4 quarts	

A pinch of coloring material such as methyl orange may be added to the mixture to enable the collector to distinguish it from other preservatives used in the field. The glycerine is a precaution against loss of the material in the event of ethanol evaporation from faulty containers. It also serves to keep the arthropod joints freely flexible. Isopropyl (rubbing) alcohol or methanol may be substituted for ethanol where the latter is not available. This preserving fluid may be used for killing, fixing, and permanently holding invertebrates having firm exoskeletons but should be used with caution in fixing soft bodied forms because of possible distortion due to dehydration.

VERTEBRATE FIXATIVE AND PRESERVATIVE

Commercial Formalin	10%	2 quarts	
Distilled Water	90%	18 quarts	= 5 gallons

This solution is commonly known as "10% Formalin" in spite of the fact that Commercial Formalin is a 40% solution of formaldehyde gas by weight in water. Fishes and the gill breathing stages of amphibians may be killed, fixed, and held indefinitely in this preservative. The chief objections to the use of this fluid are its strong acrid odor, its effect on the hands with moderate exposure and its dissolving action on the carbonates found in bones, teeth, shells, etc. (i.e. its pH is 4.0). This last objection can be overcome by the addition of buffering salts as follows:

Commercial Formalin	950 ml.
Na_2HPO_4 (Anhydrous)	93.48 gm.
$NaH_2PO_4 \cdot H_2O$	89.11 gm.
Distilled Water	Add to make 5 gallons of solution

Most of the strong odor can be removed from formalin preserved specimens if they are soaked for 20-30 minutes in the following solution before study:

$NaHSO_3$	1260 gm.
Na_2SO_3	840 gm.
Distilled Water	Add to make 5 gallons of solution

Although 10% formalin is generally used as a fixative when the "soft parts" of amy vertebrate are prepared for preservation, most research

collections of fishes, amphibians, and reptiles utilize 70-80% ethanol as a permanent preservative.

The popularity of formalin is due primarily to the fact that it is inexpensive, generally available and very effective in this capacity.

PLANT FIXATIVE AND PRESERVATIVE or TRANSEAU'S SOLUTION

This solution is named in honor of and was apparently originated by the Ohio State University plant ecologist, Dr. Edgar Transeau. Its principle advantages are the preservation of the green and other plant pigments in a near-natural state and the specimens preservation without pressing and drying. It should be pointed out that the latter technique has certain advantages such as the more efficient utilization of valuable museum space.

Commercial formalin	10%	12 quarts	
Ethanol	30%	6 quarts	= 5 gallons
Distilled Water	60%	12 quarts	

Copper Chloride or Copper Sulfate - A few crystals, enough to give the solution a faint blue-green tint.

PREPARATION OF NAIAD SHELLS FOR STUDY

The bivalve shells of the fresh-water mussels or naiads are probably as frequently found without as with the animal that produced them. It is a good practice to bring back to the laboratory a good series of both if both are available. Many of the shells collected will be found to have their features obscured by the presence of dirt in such places as the muscle scars, hinge teeth, umbonal cavity, and on the periostracum. A few moments scrubbing with an old tooth brush under a gently flowing tap of warm water is usually sufficient to remove the loose dirt. Lime deposits and/or attached algae may require soaking the specimens for several days in a strong detergent solution. It is best to allow unusually resistant deposits to remain rather than to risk damaging an otherwise valuable specimen. Once thoroughly cleaned, the shells may be placed upon toweling to drain and dry. The dry specimens are then labeled with a catalogue number or other desired data. This information should be written in India ink on the nacre of the shell and, when dry, painted over with label varnish (clear finger nail polish will do). The moderate application of olive oil with a fine brush or soft cloth will greatly reduce cracking. Excess oil may be removed with a dry cloth or by wrapping the specimens in old newspaper. Properly treated specimens have neither an oily look or feel. Care should be taken not to allow the thin shells or those having a cracked or checked periostracum to dry excessively before being oiled.

A rapid method of treating large series of specimens against cracking has been suggested by Dr. William Clench, Curator of Mollusca at The Harvard Museum of Comparative Zoology. The technique is simply the dipping of the shells into a mixture of xylene and parafin. The xylene soon evaporates leaving the specimen with a very thin protective coating of parafin. A solution of $\frac{1}{4}$ pound parafin in 1 gallon xylene has proven satisfactory.

Shell storage should be as nearly dust proof as possible and specimens may be stored for long periods of time by wrapping them along with their labels in old newspaper.

D. H. Stansbery - 1962

EXCHANGE SPECIMENS

Type labels : 1-10 specimens ..... 1 label per specimen
10-20 small specimens ..... 1 label per 2 specimens
10-20 large specimens ..... 1 label per specimen
20-200 specimens ..... 1 label per 2 specimens
over 200 specimens ..... 1 label per 3 specimens

EXCEPTION to above rule:
for very rare species, (a rare exchange species would be one that we have ≤ 1 shirtbox of) type one label for each specimen no matter how many specimens there are.

Label format :

Genus species (Author, date).
(SKIP ONE LINE)
LOCALE DATA: River, bridge, town, second town, tract + range, township, county, state, COUNTRY (in caps) if other than USA. Sec.
Collectors. collection date.
(OSUM: ___ : ___.) (field number)
ex the Ohio State Univ. Museum of Zoology.

Example:

Quadrula asperata (Lea, 1861).

Tallapoosa River at Ala. Rt. 49 bridge, at Horseshoe Bend Battleground National Park, 12.7 mi. E of Alexander City, Sec. 15, T 23 N, R 23 E, Tallapoosa Co., Alabama.
James D. Williams, 10 Oct. 1972.
ex the Ohio State Univ. Museum of Zoology.
(OSUM:1972:344).

ach species lot for exchange goes in a "small lots" box for that species. If there are enough specimens to fill an entire box, do so, and tape one extra label on the lid in the upper left corner of the end of the box.

nake sure each box goes back where you found it.

— Kathy Gail Borror
Aug. 1973

Catalogue Number	Species	Specimens s	w	d	sf	total	Collection Number	Collector	Locality: Specific	Township	County	State	Date
24725	Villosa iris iris (Lea, 1829)	s	1	0	0	1	OSUM:1970:18	John M. Condit	Big Darby Creek just W of U.S. Rt. 42 bridge, 1.1 mi. NNW of Plain City	Jerome	Union	Ohio	18 April 1970

COLLECTION RECORD CARD

I	II	♀	J	TOTAL
LS	SP	DS	SF	
s	1	0	0	1

SPECIES Villosa iris iris (Lea, 1829)

RECORDED AS

SPECIFIC LOCALE Big Darby Creek just W of U.S. Rt. 42 bridge, 1.1 mi. NNW of Plain City

TWP. Jerome CO. Union STATE Ohio

DRAINAGE → Scioto R. - Ohio R. - Mississippi R.

COLLECTED BY John M. Condit DATE 18 April 1970

IDENTIFIED BY D.H. Stansbery DATE May 1970

MUSEUM OSUM | AUTHOR

CATALOG NO. 24725 | DATE

COLLECTION NO. OSUM:1970:18 | PUBLICATION

REMARKS

FINISHED RECORD CARD USING ABOVE CATALOG INFORMATION

PROCEDURE:

LS - In this box type "s" if there is an "s" in the "s" column of the catalog. (This means that specimens were collected alive, and the soft parts removed from the shells and preserved separately in AGW.) Otherwise, leave blank.

SP - In this box type the number that appears in the "w" column of the catalog. (This is the number of specimens with soft parts, whether preserved whole or separately.)

DS - In this box type the number that appears in the "d" column of the catalog. (This is the number of specimens collected as dry shells with no soft parts.)

SF - In this box type the number that appears in the "sf" column of the catalog. (This is the number of specimens collected as dry, sub-fossil shells.)

TOTAL - In this box type the total number of speciemns. (SP + DS + SF.)

SPECIES - Copy from species data in catalog. The first (genus) name is capitalized; the other name(s) (species) are not. Include author and date as in catalog. **When lacking room, put parenthesis on second line.**

RECORDED AS - Leave this line blank.

LOCALE INFORMATION - Copy from catalog. Do not abbreviate anything unless it is abbreviated in the catalog. Directions are spelled out in full if they are parts of proper names; otherwise they may be abbreviated. (Example: "2 mi. N of East Berkshire")

DRAINAGE - This can be found on field sheet. Don't abbreviate anything except River (R.) and Creek (Cr.)

COLLECTED BY and DATE - Copy full names from catalog. If there isn't enough room here, add an * and finish up under "remarks." Take date from "date" column of catalog.

IDENTIFIED BY and DATE - Unless otherwise stated in the catalog, all specimens are identified by D.H. Stansbery at the time they are entered in the catalog.

MUSEUM - OSUM

REMARKS - Add here any additional information to be found in the catalog, such as "Donated by Herbert D. Athearn" or "From the Wheaton Collection."