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# **Procedures for Sampling Herring at the Pacific Biological Station**

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PROCEDURES FOR SAMPLING HERRING  
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ABSTRACT

Hourston, A. S., and D. C. Miller. 1979. Procedures for sampling herring at the Pacific Biological Station. Can. MS Rep. Fish. Aquat. Sci. 1554: 23 p.

Procedures for the collection, preservation and processing of herring samples are described and discussed.

Key words: Pacific herring, sampling.

RÉSUMÉ

Hourston, A. S., and D. C. Miller. 1979. Procedures for sampling herring at the Pacific Biological Station. Can. MS Rep. Fish. Aquat. Sci. 1554: 23 p.

Étude et description des procédures de collecte, de préservation et de traitement des échantillons de hareng.

Mots clés: hareng du Pacifique, échantillonnage.



## INTRODUCTION

Information from herring samples plays a critical role in annual stock assessments and forecasts (Hourston, 1979). Age determinations are the basis for assessments of year-class strength, and hence of recruitment. Data on age and weight provide a means of converting catches (in tons) to fish at age and of converting the forecast abundance of fish by year-class into tons of spawners and available catch. Length distributions show where a food fishery is most likely to capture larger fish and are used, in conjunction with information on age, to develop keys to age specimens for which suitable scales are not available and to make rough estimates of recruitment in the field. Sex ratios provide a means of monitoring the efforts of selective fishing for females (larger fish) on the composition of the stock and any differences in distribution between the sexes. Information on maturity may be related to the distribution and timing of spawnings and hence of roe fisheries. Consequently, it is important that data collected by the sampling program be as accurate and as free from bias as possible.

## COLLECTION OF SAMPLES

Samples may be collected directly from the gear, from the hold of the catcher vessel or a packer or during unloading at the plant.

In collecting samples directly from purse seine sets, care should be taken to dipnet the fish from well below the surface as small fish may be concentrated at the top of the net. Trawl or gillnet caught fish may be sampled by scooping a bucketful of fish from the catch after it has been dumped on the deck or in pens. Sampling at this stage is considered ideal as the fish have been subjected to a minimum of damage and scaling and information on the date, place and gear of capture is most accurate.

The stage of unloading at which samples are collected from the hold of a vessel (catcher or packer) does not usually affect the randomness of the sample taken (Humphreys and Hourston 1978). However, fish taken from the top of the hold before unloading has begun are preferred as they are less likely to have been damaged or scaled during transit and escape further damage during the unloading process.

Sampling in a plant should be undertaken only when it has not been possible to sample from the gear or a vessel. Such fish are usually badly scaled and are more likely to have been damaged because of the additional handling involved. Moreover, background information on the date, place and gear of capture is less reliable.

A sample may be collected in one or more scoops by a dip net or bucket. Any bias in selection should be minimized when the whole sample is collected in one scoop.

A minimum of 400 fish should be sampled from each individual fishery (Humphreys and Hourston, 1978). This involves taking four standard samples of about 100 fish. If additional samples are unlikely to be available from this location, time and gear (eg. research catches) two samples of 200 fish should be taken. On the other hand, if more samples are assured or become available, the sample size should be reduced to 50 fish. Efforts should be made to sample as many individual catches as possible spread over the entire period of the fishery, approximately in proportion to the temporal distribution of the catch.

In order to ensure adequate space-time coverage of the catch, as many as 40 samples may be required from an individual major fishery. Collectors are advised to err on the side of taking too many samples rather than too few, as samples may subsequently be culled to provide optimum space-time coverage of the catch.

#### PRESERVATION AND STORAGE OF SAMPLES

Samples should be packed in 31 cm square plastic buckets with snap-on lids available from the Pacific Biological Station or the Fisheries office at 1090 W. Pender in Vancouver, for this purpose. Alternative methods of packaging include heavy duty plastic bags securely tied or strong cardboard cartons (preferably waxed). Such containers are, however, more subject to leakage and damage during shipment.

Each sample should be securely marked with labels provided for this purpose (Fig. 1) or clearly labelled with the following information: location, date and gear of capture, name and/or CFV number of catcher boat. The sample should be preserved by freezing and kept in cold storage as much as is practicable before processing. (Transportation requirements may involve removal from a freezer for a few hours).

If freezing facilities are not available, samples may be preserved in covered plastic, metal or glass containers by salting, brining or immersion in a 5% solution of formaldehyde.

#### SELECTION OF FISH TO BE PROCESSED

When the number of samples to be processed is large, samples should be tabulated by gear, section, locality and date so that the investigator in charge can cull and assign priorities for processing.

When the number of fish in a sample does not exceed the number required for processing (50, 100, or 200 fish--see above) by more than 20%,

all the fish in the sample should be processed. When the number of fish in the sample is greater than this (eg. when 100 fish samples were taken and the requirement was subsequently reduced to 50 fish), subsampling is required. This may be accomplished by randomly scooping the approximate number of fish required from the sink or tub in which the sample was thawed. An alternate procedure is to line the fish up on a table and sample every other one on as many passes through the line as are required until the desired number of fish have been processed.

#### DOCUMENTATION

Sampling data are recorded on a standard form arrayed and coded for keypunching and subsequent EDP processing (Fig. 2). Information on the sample as a whole is presented in the top line. This includes sample number (assigned in numerical order at the time of processing); year, month and date of capture; locality number (coded from an alphabetical list of names updated annually--e.g. Hourston and Hamer 1979); CFV number (from an alphabetical list of vessel names - Anon, 1977); and gear, source, and preservation, (coded from Table 1 A-C). The length, weight, sex and maturity are recorded in that order for individual specimens in the main body of the coding sheet. Specimen data must be recorded in the order that the fish are processed so that age data may be added later by fish number. The specimen number on the coding sheet should be checked against the specimen number on the scale slide and/or otolith tray at least every 10 fish to make sure that they correspond. In the event of a mismatch, any scales or otoliths which cannot be confidently matched to the other specimen data should be discarded.

Samplers are requested to initial each coding sheet so that any follow-up inquiries may be directed to the appropriate individuals.

#### PROCESSING OF SPECIMENS

The following information is taken for each specimen in the manner indicated.

LENGTH is measured in millimeters from the tip of the snout to the end of the silvery portion of the body after the scales have been scraped clear of this area (ie. a modified standard length - Fig. 3).

WEIGHT is measured to the nearest gram, preferably on an electronic balance. Weights less than 10 grams are to be recorded to the nearest 0.1 gram. The balance should be zeroed before each measurement.

SEX is coded after Table 1E.

MATURITY is recorded on a modified ICES scale (Parrish and Savelle 1965) described in Table 2.

A SCALE is taken from the fish, preferably from the area below the insertion of the dorsal fin midway between the fin and the lateral line with forceps. If there are no scales left in this area, a reasonably large and regular shaped scale should be taken from the mid-body region anterior to the ventral fins (but not directly on the lateral line). Small, irregularly shaped and greatly thickened scales should be avoided as these are difficult or impossible to read. In badly scaled fish, such as those landed from the commercial fishery, there are usually one or more scales left under and close to the point of attachment of the pectoral fin. These scales, although irregular in shape, are usually quite satisfactory for age determinations. A scale may be cleaned by rubbing it gently between the thumb and forefinger to remove fragments of soft fragmented tissue which usually adhere on removal from the fish, and by rinsing in clean water. (Hard or vigorous rubbing or scraping may easily tear or damage the scale.) The scale is next examined for a regenerate center, which appears crystalline when the scale is held in front of a light. Such a scale cannot be aged and should be replaced immediately by a non-regenerate scale. The scale is then dipped in a mucilage solution (5-10% standard stationery mucilage in water), to which gentian violet dye may be added to make the scale more visible on the slide. Excess mucilage is then removed by shaking the scale and/or dabbing it on a paper towel. The scale is then placed on a microscope slide and any remaining excess mucilage is drawn away with forceps or a paper towel. (An excessive amount of dried mucilage blurs the pattern of the scale, especially when the dye is used.) The scales are arrayed in 2 rows of 5 on a 7.5x2.5 cm slide or 5 rows of 5 on a 7.5x5 cm slide, beginning in the upper left hand corner (fish 1), extending along the first row to the upper right hand corner (fish 5) and then beginning at the left side with the second row (fish 6), and so on (Fig. 4). The fish number on the sampling sheet should be checked against that fish number on the slide at the end of each row to make sure that they correspond (see Documentation). When the sample is completed, the slides on which the scales are mounted are labelled with the date of capture, vessel name, sample number and first and last fish number.

Other specimen data such as those given below may be required in special circumstances.

OTOLITHS may be required for age determinations when the fish are badly scaled, or for special projects. The procedure for accessing otoliths (Hourston, 1968) may be summarized as follows:

- 1) Remove the gills.
- 2) Lay the fish on its back.
- 3) Depress the gill covers laterally, exposing a triangular ridge on the underside of the skull.

- 4) Cut this ridge off level with the bottom of the skull anterior to it (Fig. 5a). This exposes, near the posterior end of the cut, a triangular bony structure pointing backwards, on either side of which is an oval opening to a deep cavity (the exposed underside of the inner ear). The otoliths are raised from these depressions with forceps (Fig. 5b). Care must be exercised in making the cut: if the cut is too deep, the brittle otoliths will be broken; if the cut is too shallow, the hole created will not be big enough to accommodate the otolith when it is grasped by the forceps. Once the otolith is dislodged from its original position, it sinks in the cavity and may be impossible to recover without considerable further dissection.

An alternative procedure for removal of the otoliths is similar to that used in sampling rockfish:

1. Lay the fish on its side, holding its head with the left hand and with the thumb under the lower opercular margin.
2. Make a scalpel cut perpendicular to the long axis of the fish, in line with the upper anterior margin of the opercular opening, and cutting primarily the top of the skull (Fig. 6a).
3. "Crack" the skull open and, without putting the fish down, hold the head bent over sharply.
4. Remove the two otoliths from their respective ampullae (Fig. 6b).

It should be noted that the sampler is trying to break through the ampullae and not to cut through them. Putting the fish down between operations greatly increases the odds that the otoliths will be displaced and not located.

On removal from the fish, the otolith is wiped clean, rinsed in fresh water and placed, with the convex, rougher side down, in the appropriate depression of an "otolith tray" (Fig. 7). These trays are panels of black plastic 14.6 cm by 9.5 cm by 0.6 cm, on one side of which there are five rows of ten round depressions 1.1 cm in diameter by 0.5 cm deep. These depressions are numbered from 1 to 50 (by rows) corresponding to the specimen number in the sample. The entire surface of this side of the tray is slightly roughened to avoid glare under a microscope. When the otoliths have been properly positioned, a few drops of 1,2 Dichloroethane ( $C_2H_4Cl_2$ ) are placed in the depression with a hypodermic needle. This fixes the otoliths to the tray. When the tray has been filled, it is labelled on masking tape applied to the bottom 2 cm of the upper side and to the bottom edge. The latter position permits ready identification when trays are filed in stacks. The label is put at the bottom of the tray to provide a convenient holding area when reading the otoliths under a microscope. Moreover, some microscopes do not have sufficient clearance between the viewing area and the post to permit observations in this area of the tray without turning it around. Trays should be labelled in pencil rather than pen as the ethanol used to clear the otoliths for reading, if spilled on the label will dissolve the ink.

GONAD LENGTH is the maximum length measured to the nearest mm. Undeveloped gonads are easily stretched and care should be taken to approximate their natural shape as closely as possible when measuring. The left gonad is from the left side of the fish (right side of the abdominal cavity when viewed posteriorly from the open, ventral side) and the right gonad is from the right side of the fish.

GONAD WEIGHT is measured to the nearest tenth of a gram on a zeroed electronic balance for the left and right gonad separately. Unless values for the gonad are specifically required, both gonads are weighed together to obtain a combined weight.

#### ACKNOWLEDGMENTS

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We also thank biologists Dennis Chalmers and Paul Sprout of the Department's Vancouver staff for their constructive comments.

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Table 1. Codings for herring sampling data.

A. GEAR		B. SOURCE	
19	Gillnet	0	Roe fishery
29	Herring seine	1	Bait fishery
20	Salmon seine	6	Food fishery
21	Other seine	2	Research - inshore
70	Beach seine	3	Research - offshore
50	Other trawl	5	Pre-fishery charter
59	Herring trawl	4	Other
01	Other		

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C. PRESERVATION		D. SEX	
0	Frozen	1	Male
1	Fresh	2	Female
2	Salted	3	Immature
4	Brine		
3	Other		

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Table 2. Maturity stages for Pacific herring sampled at the Pacific Biological Station. The descriptions of each stage are adapted from criteria developed for Atlantic herring (Bowers and Holliday 1961; Parrish and Saville 1965) and modified to suit the unique characteristics of Pacific herring which are exclusively winter-spring spawners.

Stage	State of maturity	Gonad appearance	Description	Timing
I.	Undeveloped	Thread-shaped	Virgin herring with small gonads, less than 2 mm broad. Accurate macroscopic determination of sex not usually possible.	Year-round for young herring, usually less than 150 mm in standard length.
II.	Starting	Ribbon-shaped	Gonads increased in breadth to 3-5 mm. Sex determination possible. Testes reddish-grey coloured and knife-shaped; ovaries reddish-wine coloured and rounder. (The gonads of virgins and some repeat spawners cannot be distinguished macroscopically.)	Late spring and early summer.
III.	Developing	Tube-like	Gonads thickened, increased in breadth (5-15 mm) and elongated, but not extending full length of body cavity. Ovaries red to reddish-orange with granular appearance; testes reddish-grey with smooth texture.	Late summer and early fall.
IV.	Maturing	Prominent	Gonads extend full length of body cavity. Ovaries reddish-orange to yellow; eggs distinguishable, opaque, variable in size and separable. Testes mostly grey; and will ooze sperm if sliced with a knife. Blood vessels clearly visible in the ovary and testes walls.	Late fall and early winter (slightly earlier in males than females).

Table 2 cont'd

Stage	State of maturity	Gonad appearance	Description	Timing
V.	Mature	Bulging	No blood vessels visible in gonad walls. Ovaries gold-yellow; eggs mostly transparent and uniform in size. Eggs can be exuded from the ovary under pressure and are adhesive on contact. Testes milk-white; sperm will flow under pressure.	Early winter for males, late winter for females.
VI.	Ripe	Running	Eggs transparent. Eggs and sperm flow easily without external pressure.	A few days prior to spawning (usually in late winter or early spring).
VII.	Spent	Baggy	Gonads slack. Ovaries may contain a few residual eggs. Testes limp and bloodshot.	Early spring for the first few weeks following spawning.
VIII.	Recovering	Compressed	Gonads wine-coloured and usually longer and thicker than in Stage II. Blood vessels prominent. (This stage passes into Stage III, but may resemble Stage II in the process.)	Late spring and early summer.

FISHERIES RESEARCH BOARD OF CANADA  
**BIOLOGICAL STATION**  
NANAIMO, B.C.

**HERRING SAMPLE**

CAUGHT AT.....

DATE CAUGHT.....

SEINER.....

SAMPLE TAKEN BY.....

Fig. 1. Label for herring sample.



**HERRING SAMPLE**

YEAR \_\_\_\_\_ DAY \_\_\_\_\_  
SAMPLE NO \_\_\_\_\_ BOAT \_\_\_\_\_  
AREA \_\_\_\_\_ GEAR \_\_\_\_\_  
LOCATION \_\_\_\_\_ SOURCE \_\_\_\_\_  
MONTH \_\_\_\_\_ PRESERVATION \_\_\_\_\_

INDEX	YR	SAMP NO	LOC	MON	DAY	CFV	GR	SOURCE	P
H									

INDEX	FISH NO	LENGTH	WGT	SEX	MAT	AGE
S 0 0 1						
S 0 0 2						
S 0 0 3						
S 0 0 4						
S 0 0 5						
S 0 0 6						
S 0 0 7						
S 0 0 8						
S 0 0 9						
S 0 1 0						
S 0 1 1						
S 0 1 2						
S 0 1 3						
S 0 1 4						
S 0 1 5						
S 0 1 6						
S 0 1 7						
S 0 1 8						
S 0 1 9						
S 0 2 0						
S 0 2 1						
S 0 2 2						
S 0 2 3						
S 0 2 4						
S 0 2 5						
S 0 2 6						
S 0 2 7						
S 0 2 8						
S 0 2 9						
S 0 3 0						
S 0 3 1						
S 0 3 2						
S 0 3 3						
S 0 3 4						
S 0 3 5						
S 0 3 6						
S 0 3 7						
S 0 3 8						
S 0 3 9						
S 0 4 0						
S 0 4 1						
S 0 4 2						
S 0 4 3						
S 0 4 4						
S 0 4 5						
S 0 4 6						
S 0 4 7						
S 0 4 8						
S 0 4 9						
S 0 5 0						

INDEX	FISH NO	LENGTH	WGT	SEX	MAT	AGE
S 0 5 1						
S 0 5 2						
S 0 5 3						
S 0 5 4						
S 0 5 5						
S 0 5 6						
S 0 5 7						
S 0 5 8						
S 0 5 9						
S 0 6 0						
S 0 6 1						
S 0 6 2						
S 0 6 3						
S 0 6 4						
S 0 6 5						
S 0 6 6						
S 0 6 7						
S 0 6 8						
S 0 6 9						
S 0 7 0						
S 0 7 1						
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S 0 9 3						
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S 0 9 7						
S 0 9 8						
S 0 9 9						
S 1 0 0						

Fig. 2. Herring sampling form.



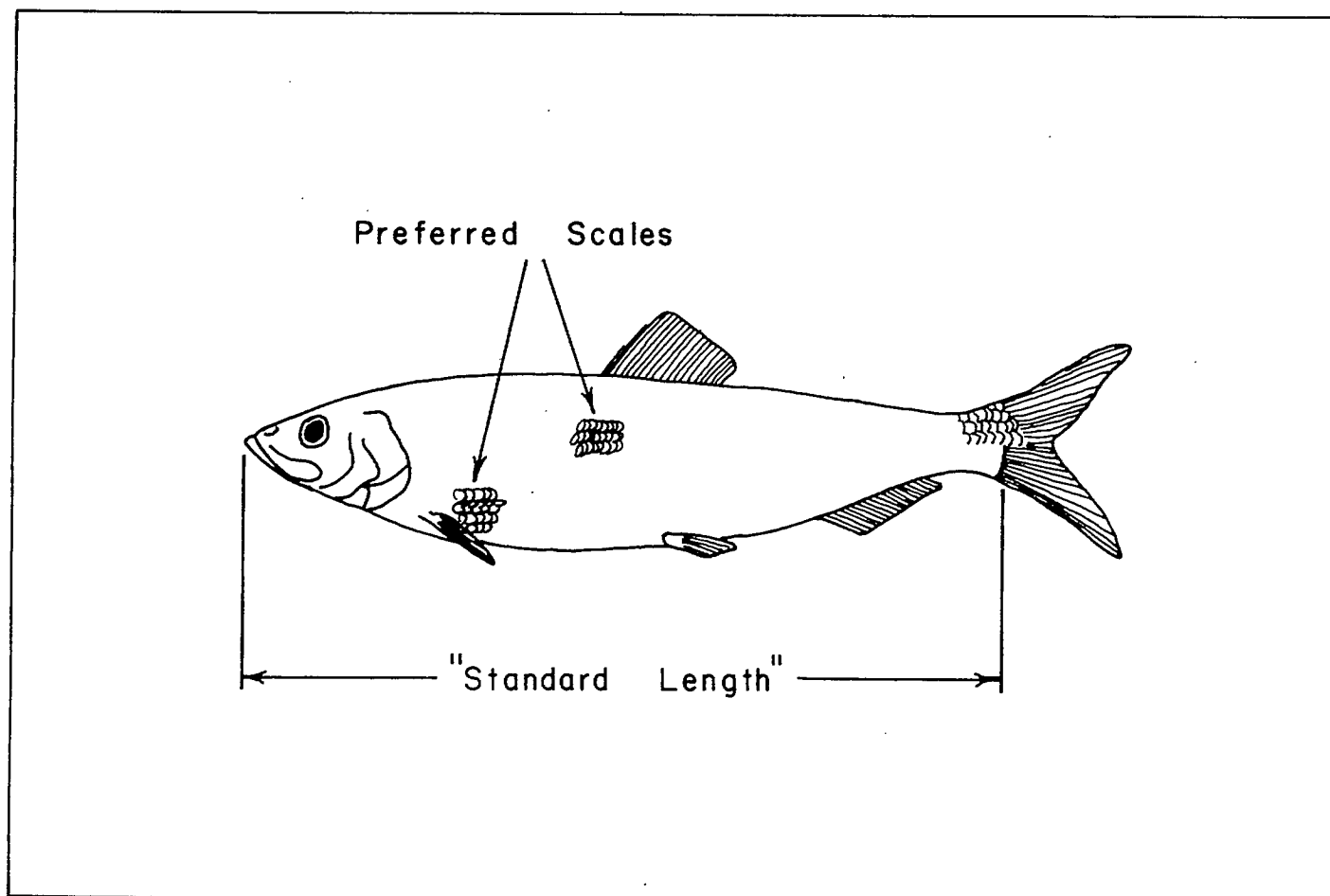


Fig. 3. Standard length as used for herring at the Pacific Biological Station and areas of preferred scales for age determination.



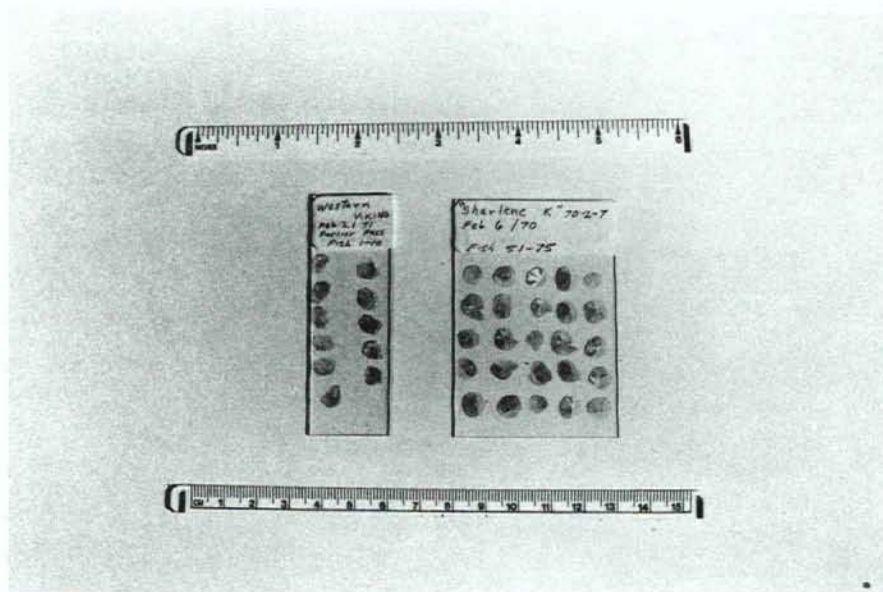
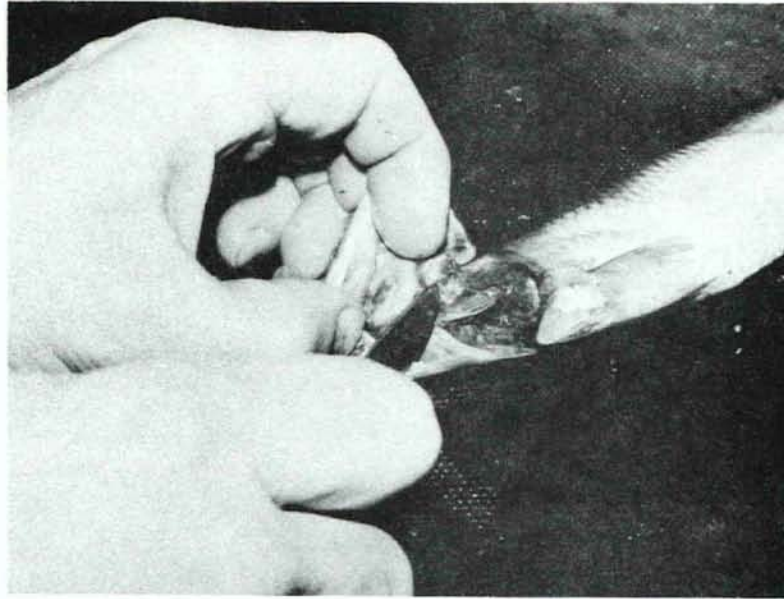


Fig. 4. 7.5 x 2.5 cm glass slide and 7.5 x 5.0 cm glass slide with mounted herring scales.



a



b

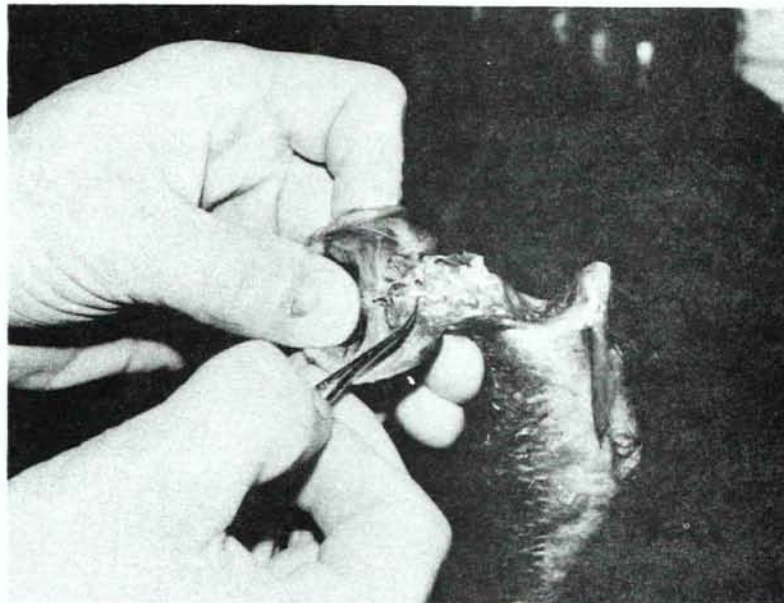
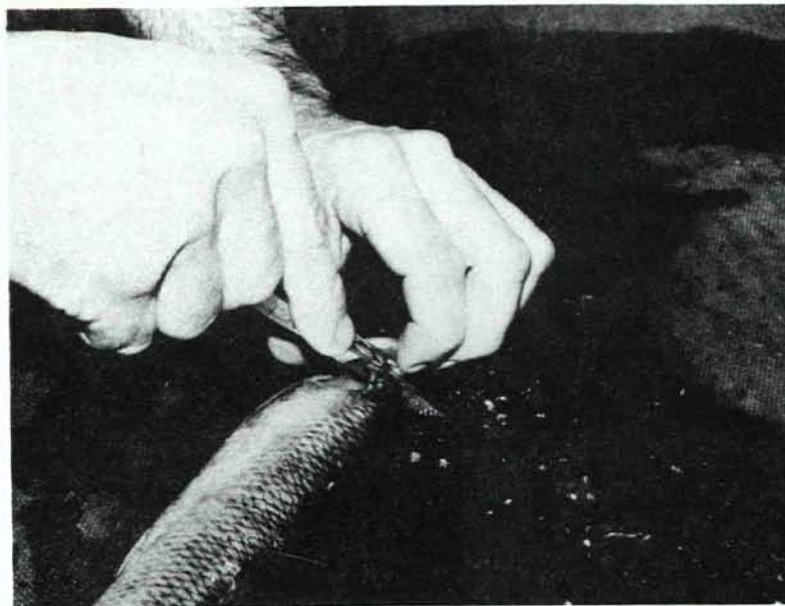


Fig. 5. The ventral approach method of otolith removal:

- a. The initial cut.
- b. Removing the otolith.



a



b



Fig. 6. The side approach method of otolith removal (rockfish method):

- a. The initial cut.
- b. Removing the otolith.



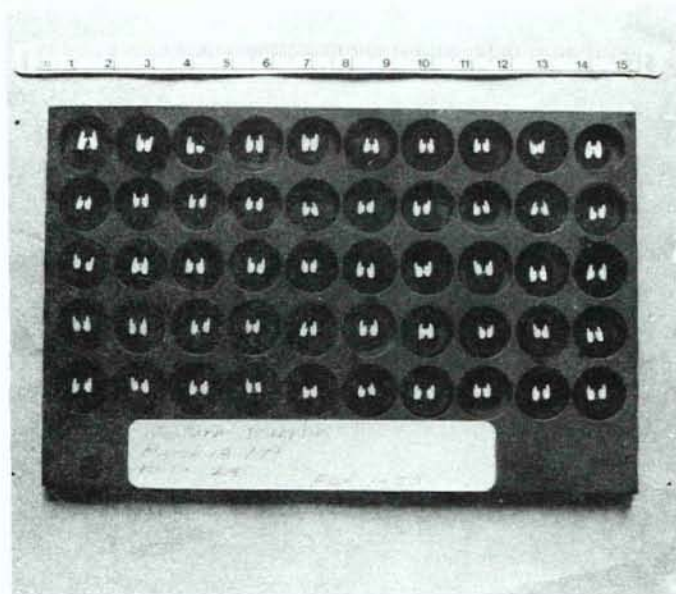


Fig. 7. Plastic otolith tray with otoliths mounted.

[illegible]

LOWE-MARTIN No. 1137

