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FIELD MANUAL FOR FISHERY BIOLOGY: METHODS FOR MEASUREMENT AND COLLECTION OF THE SAMPLES

by

Hiroyuki Yanagawa

and

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Marine Fishery Resources Development and Management Department
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INTRODUCTION

This manual describes the fundamentals of various types of examination on fishery biology in the field. This manual comprises five parts; *Measurement, Counts of Meristic Characters, Collection of Otolith and Scale, Stomach Contents and Materials for Physical Examination.*

Section on *Measurement* includes methods of measurement for fish, crab, shrimp and squid. Section on *Counts of Meristic Characters* includes methods of fin ray and scale counts. Section on *Collection of Otolith and Scale* includes collecting and storing methods. Section on *Stomach Contents* includes mainly preservation methods. Section on *Materials for Physical Examination* includes preparation and fixing the specimens. As this manual is designed for field works on fishery biology, only fundamentals on examination at the field are described. Other detailed examinations will be done at the laboratory. Detailed manual for fish biology will be published in the future by SEAFDEC/MFRDMD.

The authors would like to express their sincere thanks to Dr. Kunio Amaoka, Professor of Faculty of Fisheries, Hokkaido University, for his critical reading of the manuscript. Also, our thanks go to Mr. Ismail Taufid bin Md. Yusoff, Chief of SEAFDEC/MFRDMD, and Dr. Tamotsu Yonemori, the former Deputy Chief, for their critical comments on this manual.

I. MEASUREMENT

1. FISH

Fish shapes are divided into seven types (types A to G in *Figs. 1–1* and *1–2*) in this manual.

1.1. Total Length (TL)

1.1.1. Types A, B and D (Fish in general, tuna and tuna-like species)

The greatest dimension between the most anteriorly projecting part of the head and the farthest tip of the caudal fin.

1.1.2. Type E (Marlin and sailfish)

The maximum length between the most anterior part of the rostrum and the farthest tip of the caudal fin.

1.1.3. Type F (Sharks)

The maximum length from the most anterior part of the head to the most posterior part of the upper lobe of the caudal fin.

1.2. Fork Length (FL)

1.2.1. Types A, C and D (Fish in general, tuna and tuna-like species)

The distance from the most anteriorly projecting part of the head to the forked point of the caudal fin.

1.2.2. Type E (Marlin and sailfish)

The distance from the most anteriorly projecting part of the rostrum to the forked point of the caudal fin.

1.3. Standard Length (SL)

1.3.1. Types A, B, C and D (Fish in general, tuna and tuna-like species)

The distance from the most anteriorly projecting part of the head to the end of the vertebral column.

1.3.2. Type E (Marlin and sailfish)

The distance from the most anteriorly projecting part of the rostrum to the end of the vertebral column.

1.4. Other Parts of Types F and G

1.4.1. Type F (Shark)

Head Length (HL) is measured as the distance from the most anteriorly projecting part of the head to the last gill opening.

1.4.2. Type G (Ray)

Disc Length (DL) is measured as the distance from the most anteriorly projecting part of the disk to the posterior part of the disk.

Disc Width (DW) is measured as the distance of the widest part of the disc.

Tail Length (TAL) is measured as the distance from the mid point of the anus to the end of the tail.

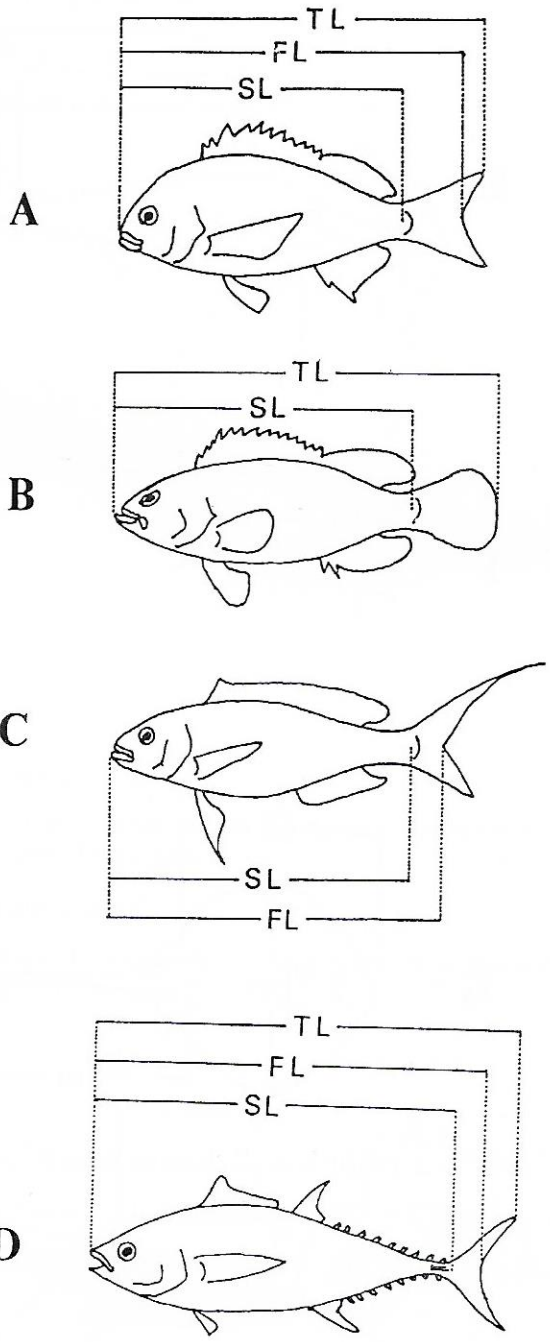
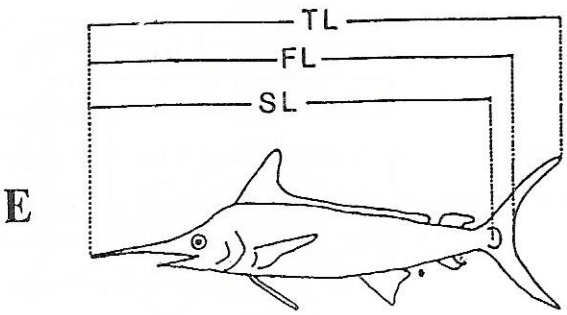


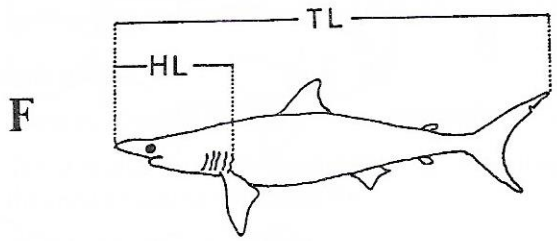
Fig. 1–1. Length measurements for types from A to D.

TL – Total Length;

FL – Fork Length;

SL – Standard Length.





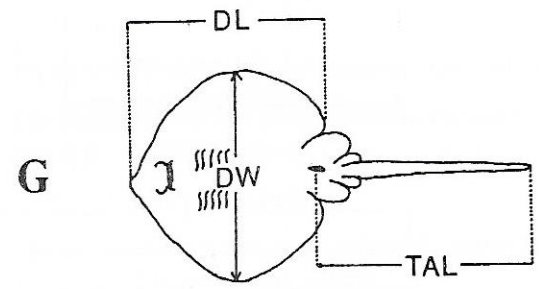


Fig. 1– 2. Length measurements for types from E to G.

TL – Total Length;

FL – Fork Length;

SL – Standard Length;

HL – Head Length;

DL – Disc Length;

DW – Disc Width;

TAL – Tail Length.

2. CRAB

2.1. Carapace Length (CL)

Carapace Length is measured as the distance from the most anterior projecting part to the most posterior part of the carapace.

2.2. Carapace Width (CW)

Carapace Width is measured as the dimension of the widest part of the carapace.

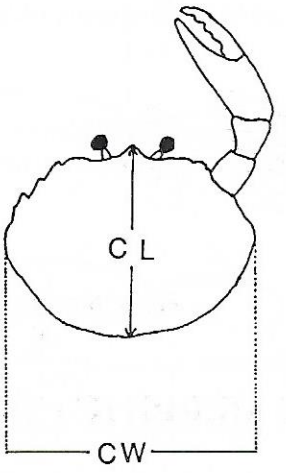


Fig. 2. Measurements for crab.
CL – Carapace Length; CW – Carapace Width.

3. SHRIMP

3.1. Carapace Length (CL)

Carapace Length is measured as the distance from the base of the eye to the posterior part of the cephalothorax.

3.2. Carapace Width (CW)

Carapace Width is measured as the dimension of the widest part of the cephalothorax from the upper view.

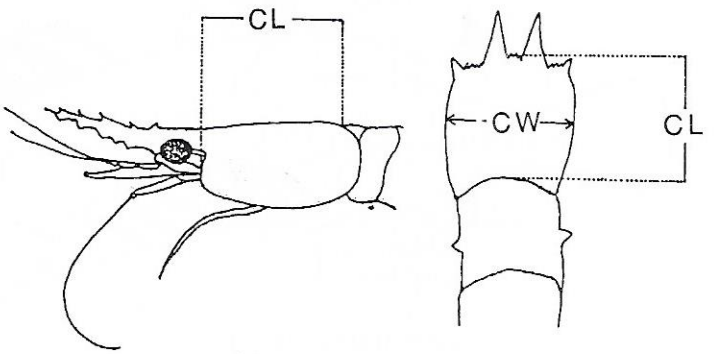


Fig. 3. Measurements for shrimp.
CL – Carapace Length; CW – Carapace Width.

4. SQUID

4.1. Mantle Length (ML)

Mantle Length is measured as the distance from the anterior edge to the posterior edge of the dorsal mantle.

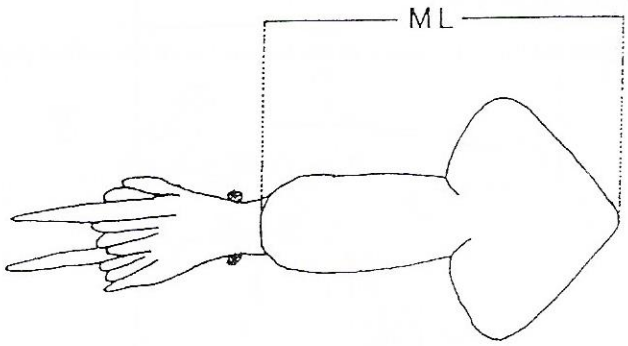


Fig. 4. Measurements for squid.
ML – Mantle Length.

II. COUNTS FOR MERISTIC CHARACTERS

1. FIN RAYS

Fin Rays are categorized into Spines (usually hard and sharp; unpaired structure and non segmented) and Rays (usually branched and flexible; paired and segmented).

1.1. Spines and Rays

Spines are designated by the Roman numerals (I, II, III .....) and Rays are designated by the Arabic numerals (1, 2, 3 .....)

In a fin containing both Spines and Rays:

1. If the two sections of the fin are separated (A), the count for the spines is separated from the Ray counts by a dash (X – 18);
2. If the two sections are conjoined (B), a comma is used to separate the counts (X, 18).

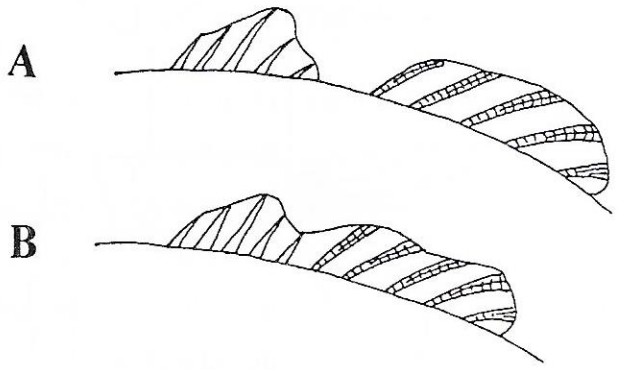


Fig. 5. Two types of fish fin.
A – Two sections of the fin are separated;
B – Two sections of the fin are conjoined.

2. SCALE

2.1. Lateral Line Scales (LLS)

Number of lateral line scales is counted as the pored scales in the lateral line or the scales along the lateral line from the shoulder girdle (refer A) to the most posterior part of the hypural bone.

2.2. Pored Lateral Line Scales (PLLS)

Number of pored lateral line scales is counted as the pored scales only along the lateral line after the shoulder girdle to the most posterior part of the hypural bone (refer B).

2.3. Longitudinal Scales (LS)

Number of longitudinal scales is counted the scales along the longitudinal raw from the shoulder girdle to the most posterior part of the hypural bone (refer C).

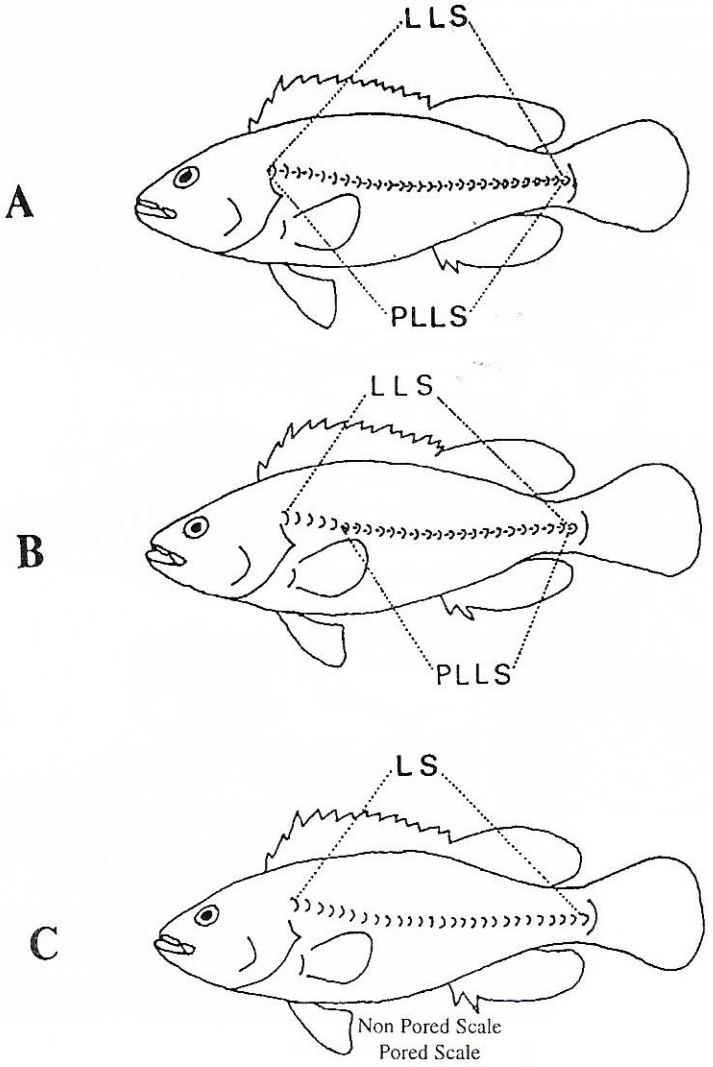


Fig. 6. Counts for fish scales.
LLS – Lateral Line Scale;
PLLS – Pored Lateral Line Scale;
LS – Longitudinal Scale.

III. COLLECTION OF OTOLITH AND SCALE

1. SCALE

The best scales for age and growth rate determination are generally to be found on the shoulder of the fish between the head and the dorsal fin *(Fig 7)*.

The fish from which scales are to be taken should be washed under cold running water. During the washing, the body of the fish should be rubbed lightly in a head-to-tail direction in order to remove any loose scales. The scales from suggested area are then removed using forceps and placed in between forefinger and thumb. Forefinger and thumb are rubbed in order to remove any mucus or tissue prior to washing with clean water.

Washed scales are normally placed on glass microscope slides and a few drops of clear water is applied before covering them with cover slips. The slides should be kept at an angle slantly to drain excess water. The scales may stick directly to the slide once they have dried up or after applying some of mounting medium at the edge of the cover glass and numbered accordingly *(Fig. 8)*. Large scales may be collected dry in small labelled envelopes for easy reference to any particular scale already recorded. Now they are ready to be observed under transmitted or reflected light.

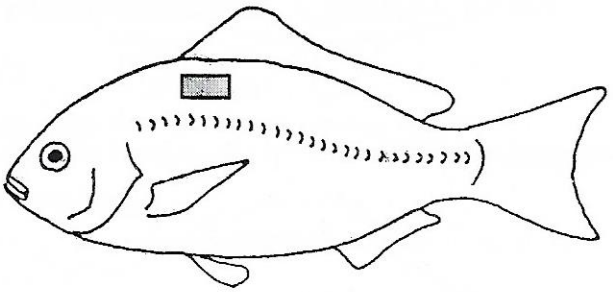


Fig. 7. Collecting position of the scales.

: Collection position

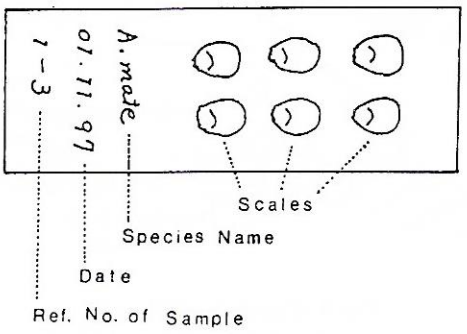


Fig. 8. Method of mounting for scales on a slide glass.

2. OTOLITH

Fish otoliths, and in particular the sagitta which are commonly the largest otoliths known to have seasonal (Christensen, 1964) and daily increments (Panella, 1971) as shown in *Fig. 9.* It is located in the sacculus of the inner ear. They are three-dimensional structures which grow at different rates in different dimensions. Like scales, otoliths vary in shapes according to fish species *(Fig 10).* A number of methods can be used to extract otoliths from any particular fish, either fresh or frozen. They are normally performed after all the measurements parameters have taken place.

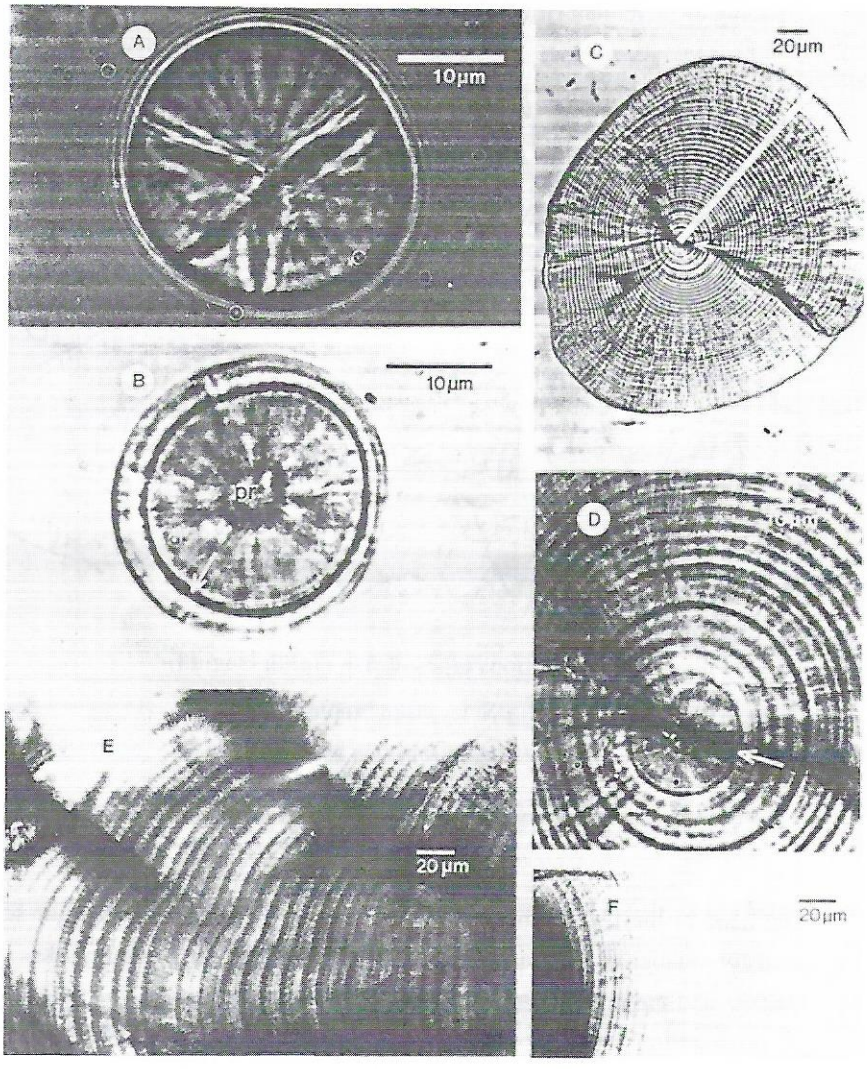


Fig. 9. Examples of otoliths with distinctive daily increment.
(Source ; Mansor, 1993)

Section of the sagitta of herring, *Clupea harengus* viewed under light microscope. *(Fig.9)*

A. A central core of the sagitta otolith from reared larval herring without increment deposition.

B. Sagitta from a reared larva at 12°C (TL = 12.9mm) showing the primodium (pr) and a heavy check marked (white arrow) of first increment.

C. Sagitta of wild juvenile (TL = 26mm) showing a heavy check mark (white arrow) used as first increment count. White line indicates the maximum posterior edge used to measure the otolith radius and for counting procedures. Anterior and posterior radii were measured from the nucleus to the most distant points before grinding.

D. Enlargement of the same otolith (C) showing the subsequent light and dark zones.

E. Sagitta showing inconsistent spacing of individual increments.

F. Spacing of increments towards otolith periphery.

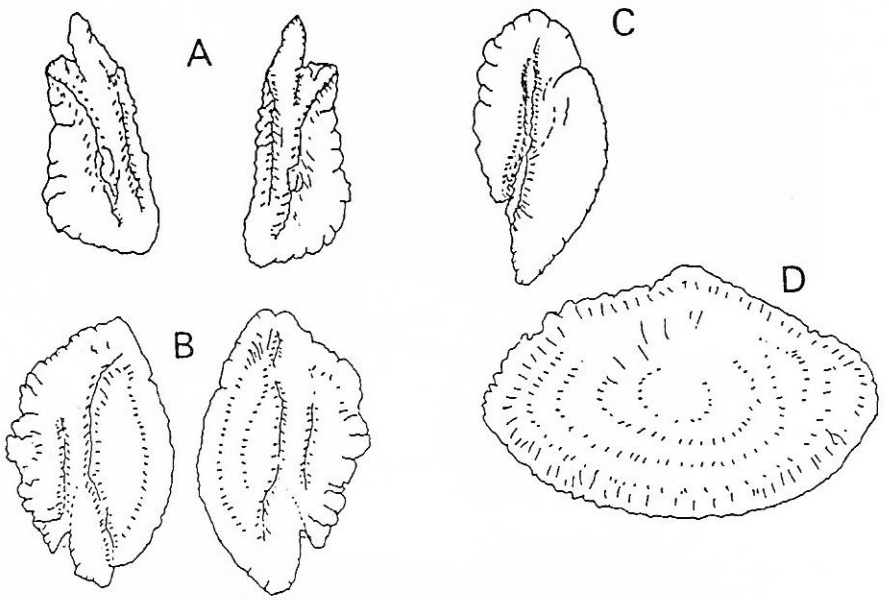


Fig. 10. Various types of fish otolith (sagitta).

A – Selar crumenophthalmus;

B – Pristipomoides multidens;

C – Decapterus russelli;

D – Lethrinus malabaricus.

The head of the fish are normally cut as shown in *Fig. 11*. A knife or scalpel can be used for smaller fish but a normal metal hacksaw for larger sizes makes the operation quicker and easier. The exposed otoliths are removed using fine forceps and cleared of surrounding tissue and washed with distilled water followed by 95% ethanol. The sagitta from larvae or post-larval fish are extracted from the head with the aid of needles and fine forceps under a dissecting microscope. They are then placed on clear microscope slides for further preparation. The right technique in cutting and removal of the otolith from the head of the fish are necessary in order to avoid damage to the otoliths.

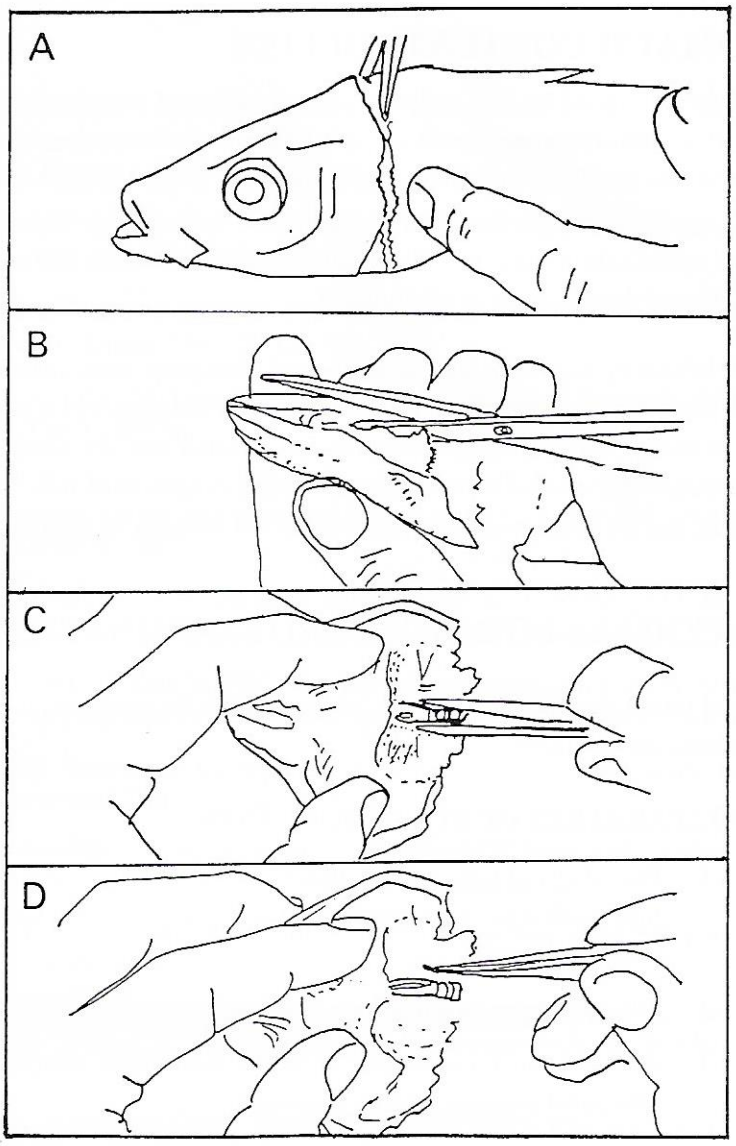


Fig. 11. Method for collecting fish otolith.
A – Fish head is cut using scissors;
B – Jaws are slit opened;
C – Sacculus is carefully broken to expose otolith;
D – Otolith is carefully removed using fine forceps.

The most simple method of storage for the majority of the fish, is to clean the otoliths thoroughly and store them dry in paper envelopes or plastic bags which are labelled and kept in boxes. The otolith may also be kept in aqueous solution. Alcohol or glycrine, an alcohol/glycerine mixture, or acetone may be used. If necessary, buffered solution should be used to ensure that the ring structure on the otolith is not damaged or rendered unreadable, especially when the daily increments are of interest.

Small otoliths are normally kept on clear microscope slides and dried in air for 24 hours or at 60°C from 30 to 60 minutes (Gjosaeter *et al.*, 1984). The dried otoliths, especially the small ones, are mounted in immersion oil under cover glass and then examined immediately under a compound microscope (Mansor, 1993). Large otoliths in need for further preparation are mounted in a hard medium such as epoxy resin onto glass microscope slides.

IV. STOMACH CONTENTS

Fish species selected for this study are normally obtained from landing ports, markets or research vessels. The stomach contents of fish samples particularly in the tropical region are digested very quickly, therefore freezing at once prior to their stomach content are taken off is very necessary to delay digestion process and bacterial activities. The other technique is to cut the ventral side or inject formalin solution (usually more than 5%) into body cavity and then preserve the specimens in 4% formalin.

In laboratory especially after completion of parameters measurement, the ventral part of fish samples are cut opened and gutted immediately and preserved in preservative solution such as alcohol for later examination. For fish of small size, the whole fish are normally preserved in 70% alcohol immediately after being caught. Each fish is measured, gutted, weighed, and the stomach contents preserved in 4% formalin for later examination.

V. MATERIALS FOR PHYSICAL EXAMINATION

Cut and take a necessary part of fish body and then fix (preserve) it in Bouin solution. Procedures are as follows.

1. PREPARATION OF BOUIN SOLUTION

1.1. Pure chemical picric acid is used for Bouin solution. Place picric acid (crystal) into distilled water for saturated solution (Ex. 10 – 15 g of picric acid and 500 ml of distilled water).

1.2. Filter the above mentioned solution (1.1).

1.3. Add 5 ml of formalin per 15 ml of picric acid saturated solution (1.2). This solution can be kept for future use.

2. FIXING THE SPECIMEN

2.1. Just before fixing the specimen, add 1 ml of glacial acetic acid into the above mentioned solution (1.3).

2.2. Put the specimen into that solution (2.1).

2.3. Rinse the fixed specimen with 70% alcohol (ethanol). The 90% alcohol can also be used for rinsing. For rinsing, it is better to prepare three or four bottles, and rinse the specimen for 30 minutes to one hour in each bottle.

2.4. Fix again the specimen in the 70% (90%) alcohol for the later examination.

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