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ABSTRACT

This manual contains a suggested Oceanographic Field Course designed as a supplement to an eighth grade science program. The three principle objectives of the course are: (1) to stimulate the interest of young students in the marine sciences; (2) to instruct students in the scientific method of field observation and laboratory investigation; and (3) to take advantage of the interdisciplinary nature of oceanography to teach the basic principles of general science. There are seven sections to the manual with suggested plans for teaching, and a suggested arrangement to accommodate the variable times in which they may be taught. Each section includes a discussion of the section topic, suggested bibliography, lesson plan, and field and laboratory procedures. Each section is concerned with sampling marine life and/or determining environmental conditions. This work was prepared under an ESEA Title III contract. [Not available in hardcopy due to marginal legibility of original document.] (HB)

TITLE III PUBLIC LAW 89-10
ESEA PROJECT

AN OCEANOGRAPHIC FIELD COURSE

FOR THE EIGHTH GRADE

BY

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The Oceanographic Education Center is a Title III Public Law 89-10 ESEA Project directed by the Town of Falmouth Public Schools in cooperation with the Woods Hole Oceanographic Institution, the U. S. Bureau of Commercial Fisheries, and the Marine Biological Laboratory.

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CONTENTS

PREFACE

The Oceanographic Education Center (OEC), an office funded under the Title III program and administered by the Falmouth, Massachusetts School system, has been seeking ways of imporving science instruction in the schools. The short field course in oceanography described in this manual is one outgrowth of that effort. It is designed to supplement the eighth grade science curriculum with projects that are not only meaningful to the students, but which will also serve as a medium for learning elementary principles of general science.

The course is taught at the Sea Farm Research Foundation property on Bourne's Pond, a typical Atlantic estuary, approximately two miles long and averaging 200 meters in width. The facilities consist of a two story chicken brooder house converted for use as a laboratory, two small skiffs, one of them equipped with a four horsepower outboard, and the estuary and surrounding marshes. The equipment is as simple as possible. Much of it is either readily available from the school system or inexpensively made from items purchased at the hardware store.

There are three prinoipal objectives of the course: (1) to stimulate the interest of young students in the marine sciences; (2) to instruct students in the scientific method of field observation and laboratory investigation; (3) to take advantage of the interdisciplinary nature of oceanography to teach basic principles of general science. The students assimilate elements of physics, biology, chemistry, and geology as an integral part of carrying out their own projects. While collecting data in the field, processing it in the laboratory, and drawing conclusions from the results, they are, as a matter course, involved in the techniques of scientific investigation.

This booklet is essentially a manual of procedures with a separate section devoted to each project. It is hoped that this arrangement will make the booklet equally useful to those who will be able to teach the course as a unit running for five consecutive school days and to those who

may want to offer it as a series of field trips scattered throughout the school year. Included with each procedure are a suggested lesson plan and description of the equipment used. The course, however, is more than a series of projects. Experimental technique is emphasized as a means of providing the students with an insight into the scientific method and an elementary understanding of the subject matter of oceanography. Some of the best opportunities for encouraging this insight and understanding are presented during the periods of casual conversation among students and teachers that occur frequently in a field course such as this. There will be plenty of time for talk during work in the field, once the particular method of collecting specimens, samples, or data has been demonstrated. To provide some guide to topics relevant to each project, there is an introductory discussion prefacing each section. These discussions are not exhaustive treatments but rather surveys of pertinent information around which the teacher can organize his thoughts about the objectives of the course, and by means of which he can guide his research. The references, listed section by section, should be consulted for thorough exposition of topics that are only touched upon here, and for definitions of unfamiliar words and technical terms that space has not allowed us to define.

The course is described as it is presently taught in Falmouth, but its content can be arranged to fit the requirements of a number of different schedules, and the procedures can be modified according to the dictates of the facilities and equipment that are available. The schedules and lesson plans are not intended as mandates that must be carried out for fear that otherwise the course would fail to achieve its objectives. The staff of the OSC is constantly modifying the course in an attempt to improve it on the basis of experience. This manual records the progress to date, and its purpose is to make it easier for others to set up an oceanographic field course for the first time in their school systems. Though marine waters are not available to all science teachers, most of the projects described here can be adapted to the study of fresh water ponds and lakes, which are accessible in many parts of the United States.

General Scheduling

The Sea Farm field course lasts for five consecutive school days. It is repeated each week for approximately ten weeks in the fall and ten weeks in the spring, depending on the weather, and is open to all eighth grade students who volunteer. The children attend in classes of not more than twenty, so that each of the almost four hundred eighth graders will have a chance to spend one week at the Sea Farm sometime during the school year.

During the week at the Sea Farm, the class is subdivided into two groups, each group of ten engaged in a different project on any given day. Two teachers are required. This arrangement provides the students with the amount of personal attention required to teach the course successfully, and insures the maximum use of the facilities and equipment.

Schedule of Activities for a Typical Week

Day	Group 1		Group 2	
1	Plankton	Teacher 1	Bathymetry	Teacher 2
2	Fish	Teacher 1	Salinity, Temperature, and Density	Teacher 2
3	Bathymetry	Teacher 2	Plankton	Teacher 1
4	Salinity, Temperature, and Density	Teacher 2	Fish	Teacher 1
5	Benthic Animals and Review	Teacher 2	Benthic Animals and Review	Teacher 1

Acknowledgments

Many people have helped to make the Sea Farm course a success. We owe special thanks to Dr. Shields Warren, president of the Sea Farm Research Foundation, Inc., who generously donated the use of his laboratory facilities, The Woods Hole Oceanographic Institution, the Marine Biological Laboratory, and the U. S. Bureau of Commercial Fisheries were also generous in lending us extra laboratory equipment and supplies.

All of the scientists of these institutions whose advice we sought were unfailingly gracious in giving up some of their valuable time to help us. We are most grateful to all of them. Though it is impossible to acknowledge each one individually, we must mention those whose time we took most frequently. Dr. Mary Sears and Dr. Melbourne R. Carriker lent us innumerable reference books and identification keys in addition to giving us valuable advice on ways to improve our experimental techniques. Dr. George D. Grice, Dr. Rudolf S. Scheltema, and Miss Johanna M. Reinhart assisted us in identifying specimens of plankton taken from Bourne's Pond.

Special thanks are also due to the administrators and teachers of the Falmouth Intermediate School, who had enough faith in the course that they were willing to have their students attend even though it meant extra work in scheduling their classes.

For help in preparing the manual, we are most grateful to Dr. Sears and Dr. W. Redwood Wright, who have read the manuscript and offered valuable suggestions for its improvement.

FISH SAMPLING AND IDENTIFICATION

Discussion

Fishes are the most noticeable water organisms because of their activity, size, and effect on other organisms. They are the predators, the voracious feeders whose influence in a biological food web may be devastating in a shallow, constricted body of water.

Fish are members of the phylum Chordata, a group of organisms possessing a complex bony or simple cartilage-like internal skeleton and a dorsal nerve cord. They are divided into three classes: (1) class Agnatha, the jawless fishes represented by the lamprey and hagfish; (2) class Chondrichthyes, the fishes such as the sharks and rays which have cartilage skeletons and small tooth-like projections or denticles all over their bodies instead of scales; and (3) class Osteichthyes, the bony fishes with true scales or flat plates covering their bodies.

These pelagic, free-swimming, organisms are widely dispersed over the oceans. However, certain species are limited to a definite area by their inability to tolerate changes or gradients in physical conditions. A single factor, or several, may limit an organism or species to that area where optimum conditions are found for that individual to breed, grow, and survive. Examples of physical factors of the marine environment which may be limiting to fish are: salinity, temperature, dissolved oxygen, the presence of toxic substances, and the absence of a particular substrate at the proper depth. Such biological factors as the number and type of predators, availability and quality of food, and territorial preference for breeding or spawning may also limit fish to a specific area. Most fishes are broadly tolerant to the preceding factors and are therefore cosmopolitan in their distribution. Fish tagged at specific sites have been recovered in all other oceans. Certain fish may undertake long migrations to search for a new food source if theirs dwindles, to escape an unfavorable environmental change, or to reach their preferred spawning ground or nursery which may be distant from the mating grounds.

Estuaries are nurseries for the young of many species of fish. The water is shallow, warm, relatively free from large predators, and rich in food. The permanent fish inhabitants of estuaries are generally small, but numerous. Hundreds of common silversides, mummichogs, sticklebacks, pipefish, and blennies dash from the shore or ripple the water when disturbed. Larger species such as striped bass, blue fish, tautog, mackerel, and mullet seasonally visit shallow water, but are seldom seen. These estuarine visitors reside in deeper, cooler water and are not as numerous. For a certain period herring alewives, eels, and salmon travel the estuaries in search of fresher or saltier waters in which to rear their young.

References

Books:

Hardy, Alister C., 1965. The Open Sea: Its Natural History. 2 parts. Boston: Houghton Mifflin Company. 335 + 322 pp.

Marshall, N. B., 1965. The Life of Fishes. Cleveland, Ohio: World Publishing Company. 402 pp.

Parker, G. H., 1936. Color Changes of Animals in Relation to Nervous Activity. Philadelphia: University of Pennsylvania Press. 74 pp.

Identification aids:

Bigelow, H. B., and W. C. Schroeder, 1953. Fishes of the Gulf of Maine. Revised edition. Washington: U. S. Department of the Interior, Fish and Wildlife Service. Fishery Bulletin 74, Vol. 53. 577 pp. (Reproduced with permission and available from the Museum of Comparative Zoology, Harvard University, Cambridge, Mass. 02138.)

Zim, H. S., and H. Shoemaker, 1956. Fishes. New York: Golden Press. 160 pp.

Lesson Plan

8:00[1] Orientation, attendance, notices.

8:10 Pick up gear from the storage barn and proceed to the field. Class members take samples for about one hour, keeping all fish in large buckets to carry back to the lab.

9:15[2] Discuss, question, answer, and lecture on (1) parts of the dissecting microscope and proper use and care of it; (2) equipment used; (3) use of the identification key, Fishes of the Gulf of Maine; (4) proper techniques of preserving organisms and handling formaldehyde; and (5) importance of fish to the ecosystem and to students.

10:30 Using the key, identify fishes collected from the estuary (usually this involves about eight different species). Include a fish dissection if time allows.

12:00 Lunch - students bring their own.

[1] This business will take longer on the first day of the week. Extra time will be needed to instruct the students in proper laboratory conduct, introudce them to any special Sea Farm rules and regulations, give them a quick tour of the facilities, and assign them to their respective groups.

[2] For each project, the teacher will need to determine the most suitable schedule for presenting lectures, discussions, and demonstrations. For instance, teachers may find that it will be effective to conduct more of the discussions and demonstrations in the field and less in the lab. To a degree, this will depend on the members of the class.

Procedures

A. Materials and Equipment

1. Dip net (see Figure 1)
2. Seine net (see Figure 2)
3. Wading boots, if necessary
4. Buckets, wash basins, or large plastic bags for transporting samples to the lab
5. Fish trap or trawl
6. Dredge net (see Figure 3)
7. Aquaria
8. 10% buffered formaldehyde solution in seawater
9. Forceps
10. Petri dishes
11. Five dissecting microscopes
12. Five dissecting kits
13. Life jackets
14. Rowboat or motor boat
15. Identification Keys

B. Method

1. Sample fish using any of the following methods:

   a. Use a dip net from a dock, a boat, or the shore. (This piece of equipment is extremely good in eel grass or muddy bottoms.) Dip the net into the water, catching some mud and/or grass. Quickly pull it out and empty it into a large container. Sort the fish out and throw the debris back.

   b. Use a seine net in shallow water (about 1 meter or less). Have two students hold the poles so the bases are on the bottom and inclined forward, then walk the seine slowly

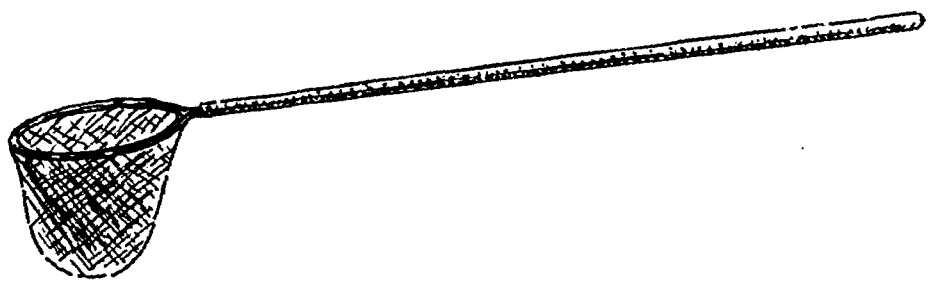


FIG. 1. DIP NET

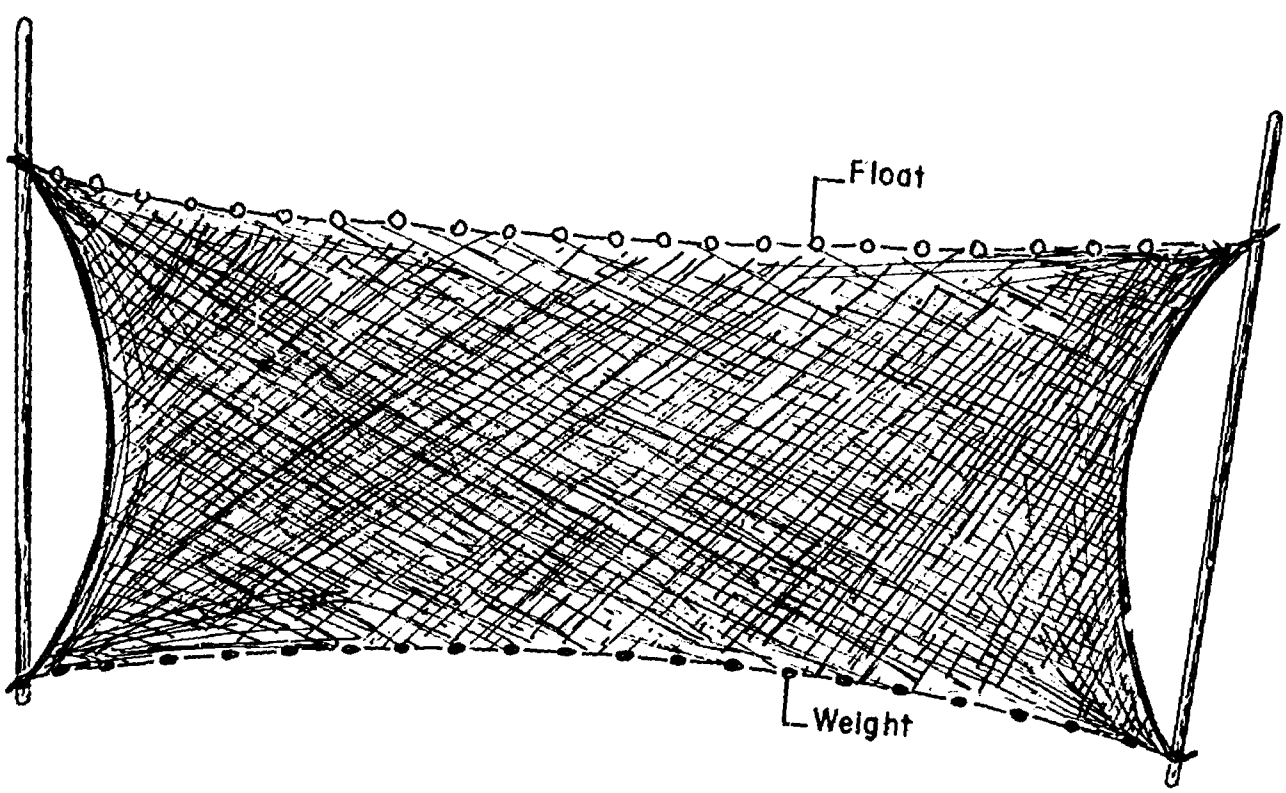


FIG. 2. SEINE NET

FIG. 3. BENTHIC DREDGE NET

through the water keeping the lead weights on the bottom and the floats on the surface. After fishing, walk the net close to shore, quickly fold the net lengthwise to enclose the fish and then lift it ashore. If there is no bank, the net may be run right onto the shore. Other methods of fishing with a seine may be devised by the teacher to fit the particular situation. (Since the students must walk with this net, it is best to use it where the substrate is firm.)

c. Use a fish trap or trawl in deeper water. Set the trap according to the manufacturer's directions. Use cracked or broken clams or mussels, rolled up bread balls, small fish, or dead fish as bait. Attach the trap to a heavy weight (a cinder block or a brick) to keep it from shifting with the current, and to a float on the surface so that it can be easily located.

d. Tow a trawl (either along the bottom or at mid-depths) behind a boat for about 10-30 minutes to ensure a good haul. After fishing, raise the trawl on deck and empty the contents into buckets.

e. Use a dredge net to capture small benthic fish.

f. Use fish traps in winter when ice covers the estuary or pond. Cut a hole in the ice, bait the trap with minnows, and set it as the manufacturer suggests.

2. Transport the fish back to the lab in buckets, large plastic pans, or large plastic bags.

3. Observe the live fish for characteristic color[3], constant movement of the gills, mucous protective covering over the body, and the action of the fins in swimming. Place some of the fish in aquaria for future observations and studies.

4. Preserve fish for identification by placing them in a closed jar containing 10% buffered formaldehyde and 90% seawater until they die.

5. Using forceps, place the fish in a petri dish and observe them with a dissecting microscope or magnifying glass.

6. Identify the fish using a key.

7. Record numbers caught, weight, length, and sex (where it can be determined easily) for each species identified. A dissection of the fish's stomach may be done to study what it had recently eaten.

[3]Sticklebacks, the common mummichog, and the common pipefish characteristically change color to match the color of their surroundings. Pipefishes can assume various shades of olive, brown, or red, and sticklebacks and mummichogs are darker in color if they are living over a dark mud substrate and paler if they are near bright sand. During the mating season, the color changes of the male mummichogs and sticklebacks are intensified. The mummichog takes on black, steel-blue, and brighter yellow markings, and the stickleback exhibits greenish and reddish hues and black dots. Mummichogs will begin to change from pale to dark in a few minutes if they are transferred from an illuminated aquarium with black walls. The reverse process also takes place. The students will be interested in discussing the reasons for this adaptation; some may want to use the microscope to examine the chromatophores which produce the change. These cells can be observed most readily in the tail of the living fish (see Parker, 1936).

PLANKTON SAMPLING AND IDENTIFICATION

Discussion

An estuarine study would be incomplete without devoting a major portion to the variable group known collectively as plankton, a word taken from Greek which means "those which are made to wander or drift." Although these plants and animals usually have some powers of locomotion, they are either so small or such feeble swimmers that they cannot make their way against the tides and currents. Most of the plankton are tiny, even microscopic, but they can be as large as the jellyfish Cyanea, which sometimes attains a diameter of over four feet. The plankton include adults and larvae from almost every phylum.

Freely floating, phtotysnthetic organ sms are called phytoplankton. These tiny plants represent the primary link in the marine food chain. They are comparable in importance to the terrestrial green plants as the converters of solar energy to usuable food material for the herbivores and carnivores. The phytoplankton reproduce by binary fission, each cell dividing to produce daughter cells; this enables rapid reproduction under favorable growing conditions, that is, when there is an adequate supply of sunlight, nutrients (principally phosphates and nitrates), carbon dioxide, and water of the proper temperature and salinity. One would expect that the phytoplankton would begin to reproduce during the months when sunlight is most intense and would gradually arrive at their maximum population sometime during the summer. This is not the case, however, In middle and high latitudes, the tiny floating plants undergo a tremendous surge in population twice a year, during the spring and the fall; during the summer months their numbers steadily decline. The sudden and spectacular increases in population, called "blooms," occur during the spring and the fall because those are the times when plant nutrients are renewed in the upper layers of the water, where photosynthesis and, therefore, reproduction can take place.

During the winter, the surface waters are cooled, become more dense, and sink while the warmer, less dense water underneath is brought to the top. The water becomes thoroughly mixed and the essential plant nutrients, instead of falling to rest on the bottom, are circulated evenly throughout the water column.[1] Consequently, there is an adequate supply in the surface waters during the spring when the light becomes intense enough for plant reproduction to begin, and the phytoplankton multiply at a great rate.

As the season progresses and the surface water becomes warmer and less dense than the underlying water, the mixing caused by winter cooling no longer takes place and a stable layer of warm water develops on the surface. As this layer becomes warm it becomes less and less likely to mix with the layer underneath and a discontinuity called the thermocline is set up. The thermocline acts as a barrier between the surface water, in which photosynthesis proceeds most rapidly, and the rest of the water column. Soon, the rapidly multiplying phytoplankton begin to deplete the supply of nutrients in these surface waters and their rate of reproduction cannot keep pace with the rate at which they are being consumed by the animals. Their numbers quickly decrease and remain at a low level throughout the summer.

In the fall, the surface waters begin to cool and sink and mixing takes place once more. If the sun is still high enough, a second bloom will occur as nutrient-rich bottom waters are circulated to the top. It is most interesting to observe the phytoplankton taken from the water during the spring and fall blooms when reproduction is so frequent that fission may be seen taking place under the microscope.[2]

[1] In lower latitudes, the principal agent of mixing is wind rather than winter cooling. Where the wind steadily blows the surface waters off shore, they will be replaced by nutirent-rich waters that well up from the depth. Upwelling is most extensive off the coasts of Chile and Peru.

[2] The process described here refers to ideal conditions in the ocean. The situation in shallow estuaries is complicated by the presence of fresh water run-offs and by the fact that variable winds are often sufficient to mix the water thoroughly from top to bottom at any time of the year.

The animals of the plankton are called zooplankton. They are herbivores and they graze on the phytoplankton, forming the second link in the marine food chain. The permanent zooplankton are those such as copepods and arrow worms that remain as plankters for their entire lives. Particular species of these will suddenly appear in the samples at a particular time of year, then will just as abruptly and regularly disappear. Other groups of marine animals, such as bivalves, gastropods, polychaetes, decapods, squids, barnacles, and fish, appear in the plankton during certain stages of their lives as eggs, larvae, and juveniles. These are known as temporary plankton and usually are found during and shortly after the peak spawning periods - spring and early fall. Jellyfish are also members of the temporary plankton. Their bell-shaped medusae are obvious in plankton tows taken during spring and summer, but they overwinter in an attached hydroid stage.

Plankton is not only vital as a live food source for other aquatic organisms, but also as decaying matter, which serves as high energy food to the mud and detritus feeders of the bottom. The tiny shells and tests of certain plankton settle to the ocean floor after the organisms have died and they become prominent constituents of the sediments known as calcareous and siliceous oozes. The calcareous oozes are the globigerina and the pteropod oozes. These sediments do not accumulate at depths below 5,000 meters because the calcium carbonate shells that predominate in them have dissolved by the time they have fallen that far through the water. The siliceous oozes are composed principally of the hard parts of diatoms and radiolarians. Their silica skeletons are not so soluble in seawater and radiolarian and diatomaceous oozes form in the deep ocean. In general, all of these sediments accumulate at the rate of 1 to 5 centimeters every thousand years.

There is much evidence that the waters over the continental shelves produce the greatest amount of organic material. Primary indications of this are the yellow or brown color of coastal waters as constrated with the blue of the open sea, and the abundance of benthic and pelagic animal life in coastal waters as compared with that of the open ocean or the abyssal depths. This great shallow water production of phytoplankton and, as a result, zooplankton makes a marine estuary the ideal place for students to develop studies of food webs and productivity.

Some additional facts of interest prepared from Johnson's (1957) treatise on plankton are:

1. The duration of the planktonic larval stage is influenced by temperature (larvae of the quahog settled in 5-7 days at a water temperature of 30°C, and in 16-24 days at 18°C).

2. To survive in the high latitudes (those near the poles), pelagic larvae have to complete their development within 1-1½ months at temperatures below 2 to 4°C. (Note: Only about 5% of Arctic invertebrates are capable of this.)

3. Much of the zooplankton undergo diurnal vertical migrations in response to light.

4. Plankton that live in tropical waters appear to require more light than those that live in the temperate zones.

5. The maximum depth to which enough sunlight penetrates for photosynthesis to occur in clear oceanic water is 150 meters at lower latitudes (those near the equator) and 30 meters at higher latitudes (those near the poles). Sunlight will not penetrate so far in estuaries, where the water is turbid.

References

Articles:

Bigelow, H. B., 1928. The plankton of the offshore waters of the Gulf of Maine. Bulletin of the Bureau of Fisheries, Vol. 40, Pt. 2, pp. 1-509.

Cushing, D. H., 1951. The vertical migration of planktonic crustacea. Biological Reviews, Cambridge Philosophical Society, Vol. 26, No. 2, pp. 158-191.

Hardy, Alister C., 1953. Some problems of pelagic life. In: Essays in Marine Biology. London: Oliver and Boyd, pp. 101-121. (Discusses vertical migration of plankton.)

Harvey, H. W., 1940. Nitrogen and phosphorus required for growth of phytoplankton. Journal of the Marine Biological Association, Vol. 24, pp. 115-123.

Johnson, M. W., 1957. Plankton. In: Hedgpeth, J. W., editor. Treatise on Marine Ecology and Paleoecology. Vol. 1, Ecology. Memoir 67, Geological Society of America, pp. 129-157.

Books:

Buchsbaum, Ralph, 1948. Animals without Backbones. Revised edition. Chicago: University of Chicago Press. 405 pp.

Fraser, J., 1962. Nature Adrift. London: G. T. Foulis and Co., Ltd. 178 pp.

Hardy, Alister C., 1965. The Open Sea. Its Natural History. 2 parts. Boston: Houghton Mifflin Co. 335 + 322 pp.

Wimpenny, R. S., 1966. The Plankton of the Sea. New York: American Elsevier Publishing Co., Inc. 426 pp.

Identification aids:

Griffith, R. E., 1961. Phytoplankton of Chesapeake Bay. Solomons, Md: Chesapeake Biological Laboratory, Maryland Department of Research and Education, Contribution No. 1720. 79 pp.

Miner, R. W., 1950. Field Book of Seashore Life. New York: G. P. Putnam's Sons. 888 pp.

Needham, J. G., and P. R. Needham, 1962. A Guide to the Study of Fresh-Water Biology. San Francisco: Holden-Day, Inc. 108 pp.

Newell, G. E., and R. C. Newell, 1963. Marine Plankton: A Practical Guide. London: Hutchinson Educational Ltd. 207 pp.

Niemeyer, V. B., and D. A. Martin, 1967. A Guide to the Identification of Marine Plants and Invertebrate Animals of Tidewater Virginia. Gloucester Point, Virginia: Virginia Institute of Marine Science, Educational Series No. 13. 82 pp.

Films:

Plankton and the Open Sea. Encyclopedia Britannica Films. 19 min., sound, color.

Leaflets:

Martek Mariner, Vol. 1 (1969), Nos. 1-6. Available from Martek Instruments Inc., 879 West 16th Street, Newport Beach, California 92660. The column "Biological Sampling" contains much useful information on plankton and plankton sampling.

Lesson Plan

8:00[3] Orientation, attendance, notices.

8:10 Pick up gear from storage barn and proceed to the field. Make two plankton tows (since all ten students can't fit in the boat at one time).

9:00[4] Discuss, question, answer, and lecture on (1) parts, care, and proper use of a compound microscope; and (2) preparation of a microscope slide. Define and discuss "plankton," "zooplankton," and "phytoplankton," and their importance to the food chain of Bourne's Pond.

10:00 Students sketch their plankters, identify them, and label with their proper scientific names. Explain how to select the proper identification key and how to use it advantageously.

12:00 Lunch - students bring their own.

12:40 Bus arrives to take students back to Intermediate School.

[3]See p. 3.

[4]See p. 3.

A. Materials and Equipment

1. Small plankton net, mesh size about 125 openings per sq. inch or 20 per sq. cm.
2. Ten compound microscopes with slides and cover slips
3. Small jars for preserving the plankton
4. Keys for the identification of the plankton
5. 5% buffered formaldehyde solution in seawater
6. Eye droppers
7. Shallow pans
8. Dissecting microscope with petri dishes
9. Motor boat or rowboat
10. Life preservers

B. Method

1. It is convenient to attach a jar at the base of a plankton net to facilitate removal of the sample (see Figure 4). If this has been done, check to be sure it is securely fastened before sampling.

2. Tow net behind boat as it is rowed or propelled slowly (about 1 MPH) by a motor across the pond for five minutes, or more. Make certain to keep the net off the bottom so that it will not tear and so that the sample will be clear and free from sand, mud, or debris.

3. If a boat is not available, students may either (a) take turns filling the net with pails of water dipped from the estuary, or (b) place the intake hose of a hand bilge pump just beneath the surface (caution, don't put it on or near the bottom) and the outake hose in the net and then pump. Using either of these methods, 10-15 minutes should be adequate sampling time.

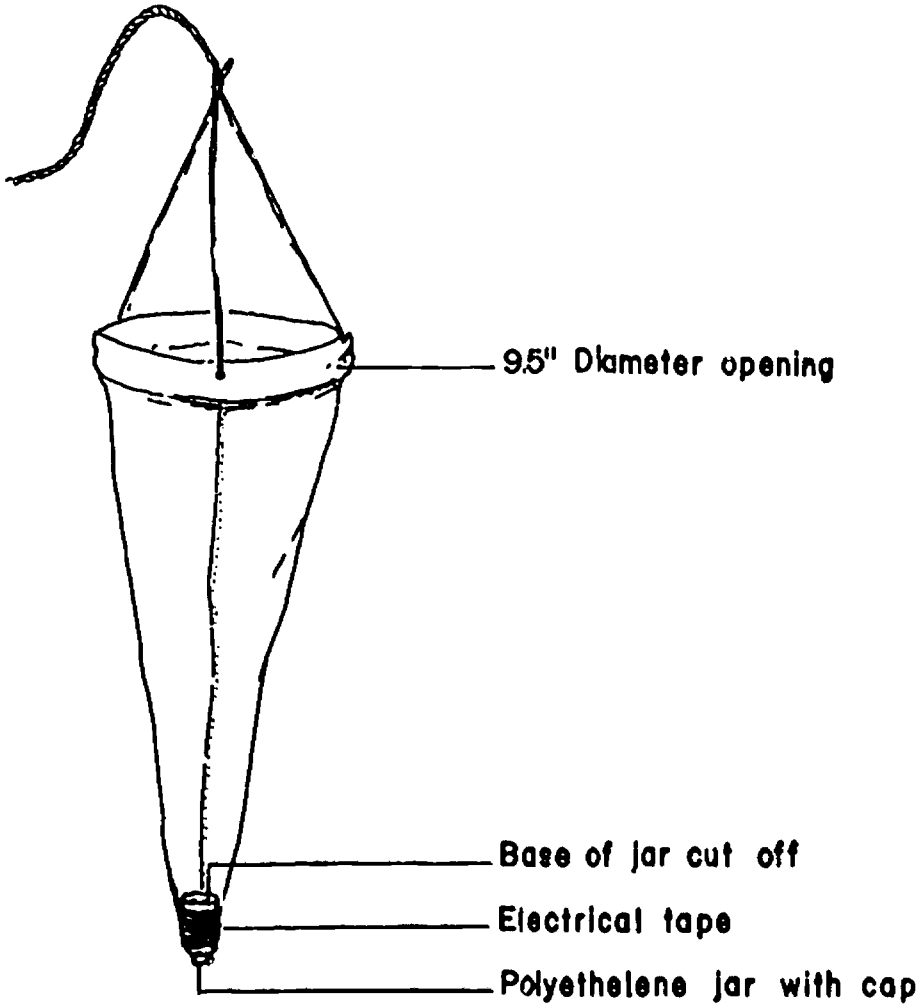


FIG. 4. STANDARD PLANKTON NET WITH END CUT OFF AND A JAR ATTACHED

4. Wash the net down with salt water to concentrate plankton in the bottom of the net or jar, and take the sample back to the laboratory.

5. Transfer the sample to a pan or wide-mouth jar from which it will be convenient for students to remove their plankton.

6. Prepare a wet slide of the living microscopic plankton and observe the plankton by using a compound microscope. (Note: Plankton will be concentrated at the bottom of the container, so be sure that students draw from the bottom of the pan with their eye droppers.)

7. Place larger plankton, such as coelentrates, crustaceans and ctenophores in a petri dish with a little seawater, then either (a) observe them under the dissecting microscope, or if one is not available, (b) view them with a magnifying glass.

8. Using the identification keys, identify as many of the organisms as possible. Note if possible what they have been feeding on.

9. Have students make drawings and keep a record of the organisms that can be identified and their frequency, or relative abundance.

10. Organisms may be preserved for future observations by placing them in a screw cap jar which is about half full with a solution that has a concentration of 5% buffered formalin and 95% seawater. Be sure to include with each drawing and each preserved organism a record of the date, the station or sampling area, and the method of sampling. These data will be essential in observing differences from tint to time and from place to place, and the number and kind of organisms captured may vary with the method of sampling.

BENTHIC SAMPLING AND IDENTIFICATION

Discussion

Biologists have separated all marine animals and plants into three broad groups: (1) the plankton, the floating organisms such as diatoms and copepods; (2) the nekton, the free-swimming or pelagic organisms such as fish, squid, and whales; and (3) the benthos, the bottom-dwelling animals such as clams, worms, and starfish. Benthic organisms are an interesting and varied group. Sand dollars, hermit crabs, snails, sponges, hydroids, and even some fish creep and crawl along the ocean bottom or burrow into the substrate. Some, such as corals or sea weeds, remain firmly anchored to the bottom. The benthos has been subdivided into two groups: (1) the epifauna, comprising all the animals and plants which remain on the surface of the substrate; and (2) the infauna, including all the animals whose habitat lies below the surface of the substrate.

According to Thorson (1957), the epifauna occupies on an average less than 10% of the total area of the sea bottom and reaches its greatest numbers in very shallow water. These organisms must be adapted to meet the rapid changes in temperature, salinity, nutrients, and dissolved oxygen that are inherent in shallow waters. When intertidal epifauna are stranded in tidal pools after the tide recedes, they must withstand the drying, hot rays of the sun and the sharp increase in the salinity of the pool that results from evaporation. Conversely, they must survive the drop in salinity of the water after a rain, and the freeze of winter. Since only very tolerant organisms can survive this ever changing environment, the number of different species is low. Those species that are adaptable to the shallows find food and space to be plentiful. As a result they have rapid growth, high survival of young, and a general increase in population. Predators that can survive this fluctuating environment are also few in species, but high in numbers. Such predators as sea stars, brittle stars, and gastropods take a heavy toll of shellfish.

The infauna, by the very fact that they burrow beneath the surface of the substrate, are not exposed to such rapid changes in temperatures as are the epifauna. Due to their inaccessibility, they are also less likely to be the diet of predators. Thus, it seems that both the numbers of species and the populations should be high. Thorson (1957), however, states that although the infauna may occupy more than half the surface of our globe, the total number of species is only about one fourth that of the epifauna. Although environmental conditions are ideal, only a small number of species have adapted to this subsurface life. Infaunal organisms are most prevalent below the intertidal zone and are usually associated with level bottoms.

Only the Arctic and Antarctic coastal waters and the very deep sea are inhabited by stable benthic communities in which the adults inhabit the same area for long periods of time without being disturbed by invaders. The constancy of such major physical factors as temperature and salinity has produced relatively stable environments that allow established communities to remain for such long periods. These inhabitants are generally known to be longer lived and reach larger sizes than their counterparts in warmer waters. Their young generally remain close to the community because they are either born live, brooded by a parent, or have a very short pelagic larval period.

In contrast, 85-90% of all tropical benthic species have a long pelagic life. Likewise, in the cold temperate seas about two thirds of the benthic animals have long lived pelagic larvae (Thorson, 1957). This is a disadvantage to the community, since larvae are especially vulnerable to predators and changes in temperature, salinity, and light. They may even be washed from the boundaries of the community by storms or strong currents. In these waters the physical environment changes rapidly, vast numbers of species compete for available space, and predators abound. With such selective pressures, new communities continuously encroach upon and finally succeed established communities. These communities, then, are highly unstable and difficult to define and study.

An outside factor which may drastically alter the structure of the benthic community in coastal waters is the domestic and industrial waste which is increasingly dumped into streams and bays. Massive crab kills, the disappearance of many species, and changes in spawning habits have been linked to pollutants. Specific pollutants which have been cited as adversely affecting aquatic life are pesticides and sewage, which can sometimes seep down into the ground water and run out into estuaries even when not placed there directly. Thermonuclear power plants, chemical plants, and various mills which expel their effluents into the coastal waterways are sources of pollution. In certain situations, heated water alone can be a pollutant. Thermonuclear power plants located along the shore can discharge vast quantities of hot water from their cooling systems into the waterways; enough to alter the aquatic environment to the point that shellfish and other bottom-dwelling organisms native to the area cannot survive.

Another hazard to benthic populations is disease. Any infectious disease which is introduced to a close-knit community spreads throughout the entire community as an epidemic. Recently, the oysters of Chesapeake Bay have been infected with a fatal disease that has practically halted the vast oyster industry there. The wasting disease of eel grass which appeared along the Virginia shore in 1931 spread rapidly up and down the coast and was even carried across the Atlantic. Within two years, the infectious parasite which caused the disease had almost entirely destroyed the plants along the Atlantic coasts of both Europe and North America. The animals that live in close association with eel grass also disappeared; among them, clams, oysters, and bay scallops. In many places, it was twenty years before the eel grass communities began to flourish again.

In addition to pollution and disease, overfishing is a severe hazard to commercial benthic species. Because of their ease of capture and their seemingly endless supply, edible shellfish along the western Atlantic shore have been heavily fished since the time of the Indians. As a result, their numbers have been so far reduced that the federal government and the coastal states have had to impose restrictions on shell-

fishing. Now scientists and commercial companies study the problems of these benthic populations in an attempt to find ways of regenerating the depleted species.

References

Articles:

Clark, John R., 1969. Thermal pollution and aquatic life. Scientific American, Vol. 220, No. 3, 18-27.

Coe, W. R., 1912. Echinoderms of Connecticut. Connecticut State Geological and Natural History Survey, Bulletin 19. 152 pp.

Hedgpeth, J. W., 1954. Bottom communities in the Gulf of Mexico. U. S. Fish and Wildlife Service, Fishery Bulletin 89, Vol. 55, pp. 203-214.

Jones, N. S., 1950. Marine bottom communities. Biological Reviews, Vol. 25, No. 3, pp. 283-313.

MacGinitie, G. E., 1939. Littoral marine communities. American Midland Naturalist, January, Vol. 21, No. 1, pp. 28-55.

MacGinitie, G. E., 1940. Life in a marine mud flat. Turtox News, Vol. 18, No. 1, pp. 1-3.

Milne, Lorus J. and Margery J., 1951. The eelgrass catastrophe. Scientific American, Vol. 184, No. 1, 52-55.

Swan, E. F., 1962. The growth of the clam Mya arenaria as affected by the substratum. Ecology, Vol. 33, No. 4, pp. 530-534.

Thorson, G., 1957. Bottom communities. In: Hedgpeth, J. W., editor. Treatise on Marine Ecology and Paleoecology. Vol. 1, Ecology. Memoir 67, Geological Society of America, pp. 461-534.

Yonge, C. M., 1948. Bottom fauna of the sea. Research, Vol. 1, No. 13, pp. 589-595.

Books:

Buchsbaum, Ralph, 1948. Animals without Backbones. Revised edition. Chicago: University of Chicago Press. 405 pp.

Clarke, George L., 1965. Elements of Ecology. Revised edition. New York: John Wiley & Sons, Inc. 560 pp.

Day, J. H., 1969. A Guide to Marine Life on South African Shores. Cape Town: A. A. Balkema for the University of Cape Town. (Includes valuable information for the amateur about sampling and collecting techniques and the effects of environmental factors such as waves, tides, currents, and salinity on shallow water fauna.)

Hedgpeth, Joel W., editor, 1957. Treatise on Marine Ecology and Paleoecology. Vol. 1, Ecology. Memoir 67, Geological Society of America. 1296 pp.

Sverdrup, H. V., M. W. Johnson, and R. H. Fleming, 1946. The Oceans: Their Physics, Chemistry and General Biology. Englewood Cliffs, N. J.: Prentice-Hall, Inc. 1087 pp.

Turner, H. J., 1949. Report on Investigations of Methods of Improving the Shellfish Resources of Massachusetts. Department of Conservation, Division of Fisheries in Massachusetts, prepared by the Woods Hole Oceanographic Institution. 22 pp.

Identification aids:

Needham, J. G., and P. R. Needham, 1962. A Guide to the Study of Fresh-Water Biology. San Francisco: Holden-Day, Inc. 108 pp.

Niemeyer, V. B., and D. A. Martin, 1967. A Guide to the Identification of the Marine Plants and Invertebrate Animals of Tidewater Virginia. Gloucester Point, Virginia: Virginia Institute of Marine Science, Educational Series No. 13. 82 pp.

Miner, R. W., 1950. Field Book of Seashore Life. New York: G. P. Putnam's Sons. 888 pp.

Smith, R. I., editor, 1964. Keys to Marine Invertebrates of the Woods Hole Region. Systematics-Ecology Program, Marine Biological Laboratory, Contribution 11. 208 pp.

Zim, H. S., and L. Ingle, 1955. Seashores. New York: Golden Press. 160 pp.

Films:

Life in the Sea. Encyclopedia Britannica Films. 11 min., sound, color.

Seashore Life. Encyclopedia Britannica Films. 11 min., sound, color.

Lesson Plan

8:00[1] Orientation, attendance, notices.

8:10[2] Pick up gear from storage area and proceed to the shore. Sample the shore infauna and place specimens from different substrates in different pans. Sample the epifauna and keep the organisms from this habitat separate. Carry specimens and gear back to the lab. Discuss and define these micro-environments while in the field.

10:00 Review the use of the dissecting microscope and discuss how to use benthic identification keys. Discuss classification of organisms.

10:30 Identify benthic samples using proper biological classification.

12:00 Lunch - students bring their own.

12:40 Bus arrives to take students to Intermediate School

[1]See p. 3.

[2]See p. 3.

Procedures

A. Materials and Equipment

1. Two sieves, one with large-mesh screen and one with small
2. Shovel and/or scoop
3. Two shallow pans
4. Dredge net (see Figure 3, following p. 4)
5. Glass-bottomed box or face mask
6. Rowboat or motor boat
7. Life jackets
8. Buffered formaldhyde solution, 10% in seawater
9. Forceps and dissecting needles
10. Petri dishes
11. Five dissecting microscopes
12. Invertebrate keys or identification guides

B. Method

1. View benthic fauna in a natural state with the mask or glass-bottomed box and make notes on observations. Be sure to note type of bottom and vegetation.

2. Sample the infauna by digging along the shore with a shovel and placing the sample in the sampling screen, being sure the larger mesh fits snugly inside the smaller one. Shake the two screens back and forth so that the sample lies just below the water. (Caution, be sure the sieves are not completely inundated or the sample will be lost.) The moving water will separate the sample according to size. The large specimens and debris will remain on the top screen, the small specimens and particles of sand and shell will lie in the lower screen, and the silt and find sand will sift through both screens. This technique may be used for sampling shallow water infauna by very carefully

shoveling or scooping the substrate and some water into the screens and shaking them as outlined above. Transfer the separated sample to shallow pans for transportation back to the lab.

3. Sample the epifauna by dragging the dredge net along the bottom. This may be done from a slowly moving boat, or the students may tow the dredge by hand as they walk along the shore. Remove the specimens from the net and place them in shallow pans. Transport all specimens back to the lab.

4. Observe the living specimens under a dissecting microscope. Note color, size, definitive characteristics, and method and efficiency of locomotion. Place specimens to be identified in killing jars containing a 10% solution of buffered formaldehyde in seawater.

5. Identify specimens completely using biological classification (Kingdom, Phylum, Class, Order, Family, Genus, and Species). Sketch specimens. Label sketches and preserved specimens with genus and, when possible, species names, date, station or sampling area, and method of sampling.

BATHYMETRY

Discussion

Bathymetry is distinguished from other branches of oceanography in that data have been collected over centuries rather than decades. As long as ships have sailed it has been the practice to sound the depth of the water with a weighted line to keep from running aground on hidden rocks and shoals. From the sixteenth century, when European navigators began to feel their way along the shores of newly discovered lands, soundings began increasingly to be incorporated into charts and maps of coastal waters. By the beginning of the nineteenth century, the perimeter of the North Atlantic and the configuration of the floor beneath its coastal waters was pretty well charted. However, with the exception of perhaps a hundred soundings of doubtful accuracy, no measurements of the deep ocean floor had been made. It had been assumed that the ocean basins were flat and featureless plains shaped much like a soup bowl gradually and continuously deepening towards the center.

Systematic deep sea soundings were first made during the middle of the nineteenth century as a means of finding suitable routes for laying submarine cables. The first contour map of an entire ocean basin, the North Atlantic, was published by Matthew Fontaine Maury, an American hydrographer, in 1854.

About this time, scientific interest in the oceans had been aroused. Necessary for studies of any kind, geological, physical, or biological, was a knowledge of the shape of the ocean basins. By 1886, Sir John Murray, director of the Challenger Office, was able to make maps of the Atlantic, the Pacific, and the Indian Ocean basins based on approximately 6,000 soundings. In 1904, the International Geographical Congress produced charts of all the ocean basins based on about 18,000 soundings. With this number of soundings, the existence of major relief features on

the sea floor, ridges and undersea mountains, began to be revealed providing useful information for scientists who were interested in the structure and history of the earth.

The technique of echo sounding was introduced during the 1920's and it became useful enough to replace the laborious method of lead line sounding by the 1930's. This innovation, together with increasingly precise methods of navigation, has made it possible to construct maps of the sea floor with vastly improved accuracy and detail. Though there are still large areas of the earth's underwater surface that are sparsely charted, the characteristic topography of the sea floor is known in great detail and we have a clear picture of the continental shelves, slopes, and rises, of the global system of oceanic ridges, the abyssal plains, and the rugged mountainous terrain.

References

Articles:

Burmister, C. A., 1957. Electronics in hydrographic surveying. 1. How deep is the ocean? International Hydrographic Review, Vol. 34, No. 2, 117-125.

Books:

Adams, K. T., 1942. Hydrographic Manual. Revised edition. U. S. Coast and Geodetic Survey, Special Publication No. 143. 940 pp.

Kjellstrom, Bjorn, 1967. Be Expert with Map and Compass: The "Orienteering" Handbook. Revised edition. La Porte, Indiana; American Orienteering Service. 136 pp.

Richards, G. S., 1967. The Admiralty Chart: British Naval Hydrography in the Nineteenth Century. London: Hollis and Carter.

Maps and charts:

Heezen, Bruce C., and Marie Tharp. Physiographic Diagrams of the South Atlantic, the North Atlantic and the Indian Oceans. Geological Society of America, 419 W. 117th Street, New York, New York 10017.

The navigational charts issued by the U. S. Coast and Geodetic Survey can be obtained from local boating or fishing supply stores.

Lesson Plan

8:00[1] Orientation, attendance, notices.

8:10[2] Discuss, question, answer, and lecture on (1) the definition of bathymetry; (2) why it is useful; (3) what a sounding is and how to make it; (4) locating and mapping positions; and (5) the metric system.

8:30 Pick up gear from the lab and the storage barn and proceed to the field. Discuss and demonstrate the use of a magnetic compass, the meaning of magnetic north and true north, the meaning of a map scale, and ratio and proportion. Students determine and map the positions of profile lines along which soundings will be made. Read the tide gauge.

10:30 From the boat, demonstrate the technique of taking soundings with a lead line. Students take soundings and record them.

11:15 Return to the laboratory where students complete their topographic maps using the information they gathered in the field.

12:00 Lunch - students bring their own.

12:40 Bus arrives to take students back to the Intermediate School.

[1]See p. 3.

[2]See p. 3.

Procedures

A. Materials and Equipment

1. Map of the estuary and surrounding area (an official town map, U. S. Geological Survey map, etc.); 10 copies
2. Work sheet (Figure 10); 10 copies
3. Ten writing boards or clipboards
4. Wooden tripod stand with moveable legs (for convenience, a 12" x 12" plywood working table should be mounted at the top)
5. Magnetic compasses
6. Protractors
7. Parallel ruler
8. Metric rulers
9. Pencils
10. Measuring line, $\frac{1}{4}$" nylon, 10 meters long, graduated in decimeters
11. Lead line, 4 meters long, graduated in decimeters (Figure 5)
12. "Marks-a-lots" (for calibrating measuring line and lead line)
13. Meter stick
14. Crayons, five different colors
15. Tide gauge (Figure 6)
16. Field tote box
17. Rowboat or motor boat
18. Life jackets
19. Stopwatch, or a watch with a sweep second hand
20. Range finder, if desired

B. Method

1. Assemble all field equipment (tripod, maps, pencils, protractors, compasses, parallel ruler, metric rulers, 4 meter lead line, 10 meter measuring line, stopwatch, life jackets, and outboard motor) and proceed to the estuary. Each student should have a copy of the work sheet and the map taped to a writing board or

clipboard of convenient size. Near the area of the estuary to be mapped locate a permanent landmark such as A (as illustrated in Figure 7), a building that is already shown on a copy of the map that is also clearly visible from the water. Set up surveying equipment at the most convenient corner of the building. Using the protractor, have the students construct a magnetic north line, B, on their maps. (In Falmouth, the mean declination is 14.5° W of N.). Using the compass, have the students take a bearing on a visible landmark, C, across the water. Make a note of the angle. Using a parallel ruler, have each student construct the angle of the bearing, thereby establishing the first profile line, E, on his map. Working from the same point, establish addtional profile lines in a similar manner, making sure that there are enough to give adequate coverage of the estuary to be mapped.[3]

2. On their work sheets, have the students record the level of the water as indicated on the tide gauge.

3. Assemble the necessary equipment (the work sheets and the maps, the lead line, pencils, and life jackets) and proceed to the

[3]For some sections of the estuary there will be no landmarks already shown on the map that will be visible from the water. In order to map those sections it will first be necessary for the class to establish such a landmark by the following method. Select some outstanding feature of the landscape, such as a telephone pole or a conspicuous tree trunk, that is clearly visible both from the water and from some position already shown on the map. Working from the known position and following the procedure for establishing profile lines on the map, construct a sight line that runs from the known position in the direction of the telephone pole. Using the 10 meter line, measure the distance between the two points along the direction of sight. Convert this distance to scale and, using the metric ruler, mark the position of the telephone pole on the map. Establish profile lines from this point as described above.

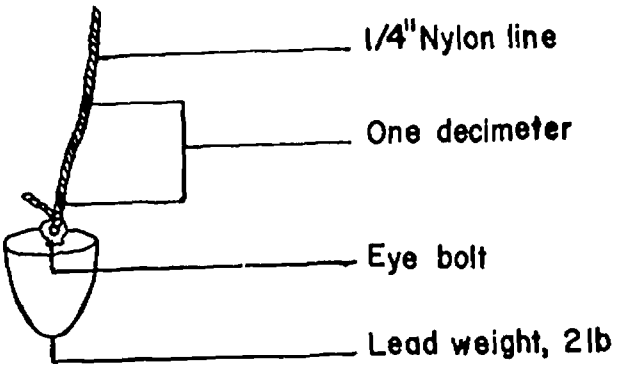


FIG. 5. LEAD LINE

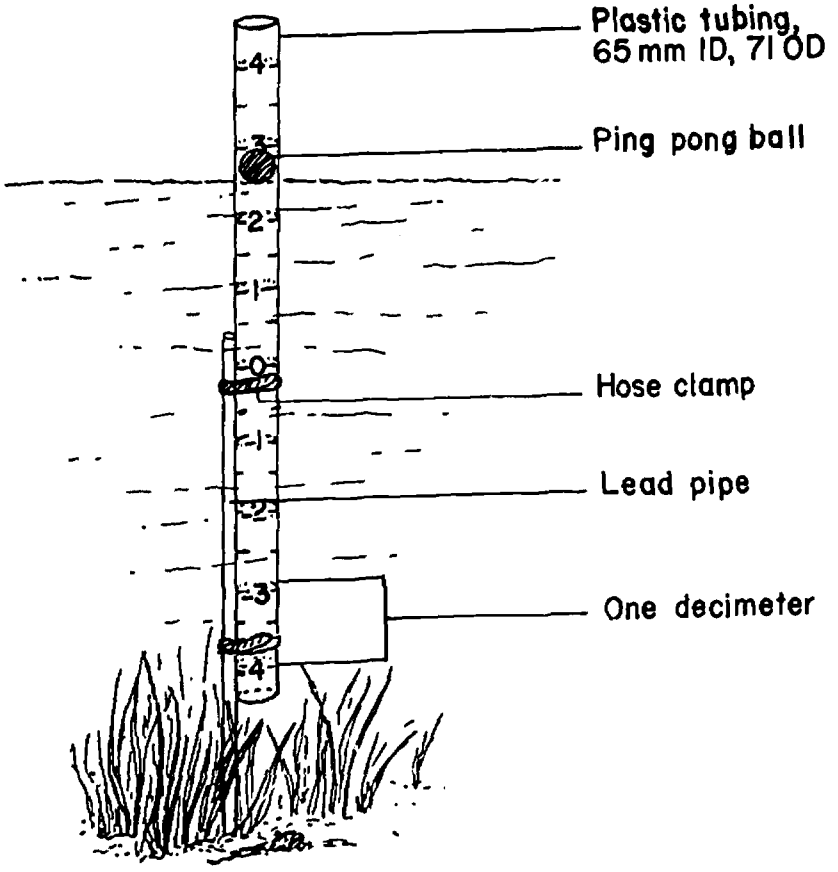


FIG 6. TIDE GUAGE

Official Map

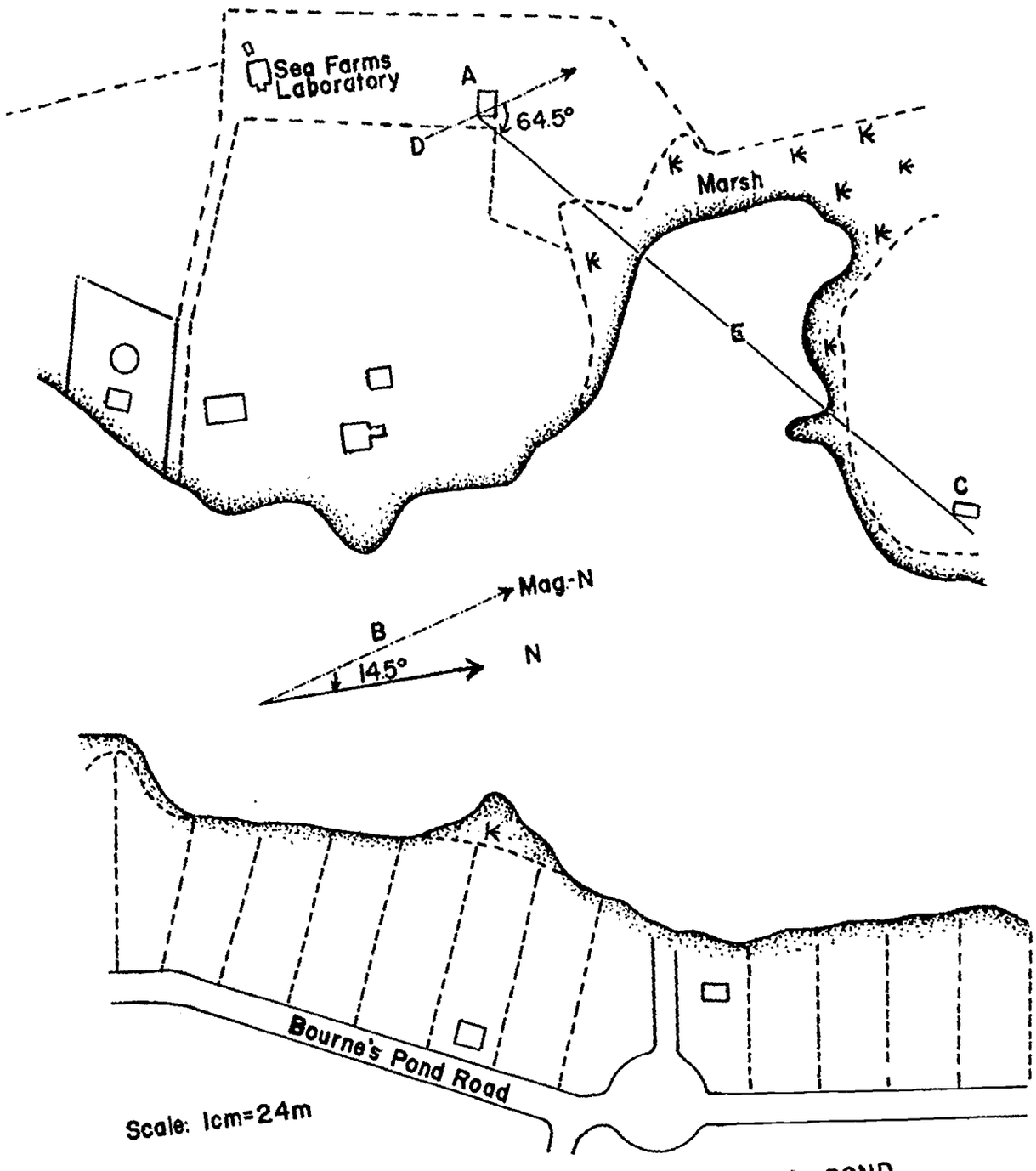


FIG.7. MAP OF A PORTION OF BOURNE'S POND

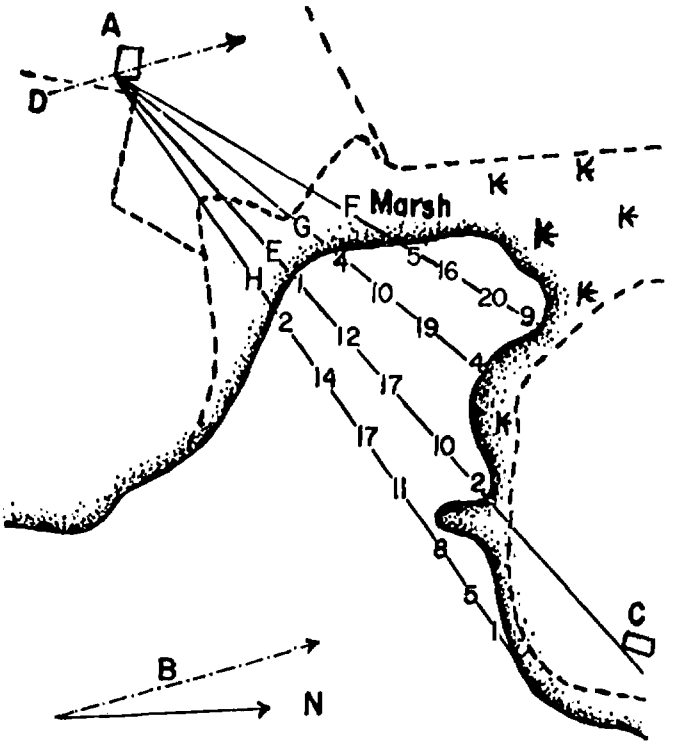


FIG. 8. PROFILE LINES

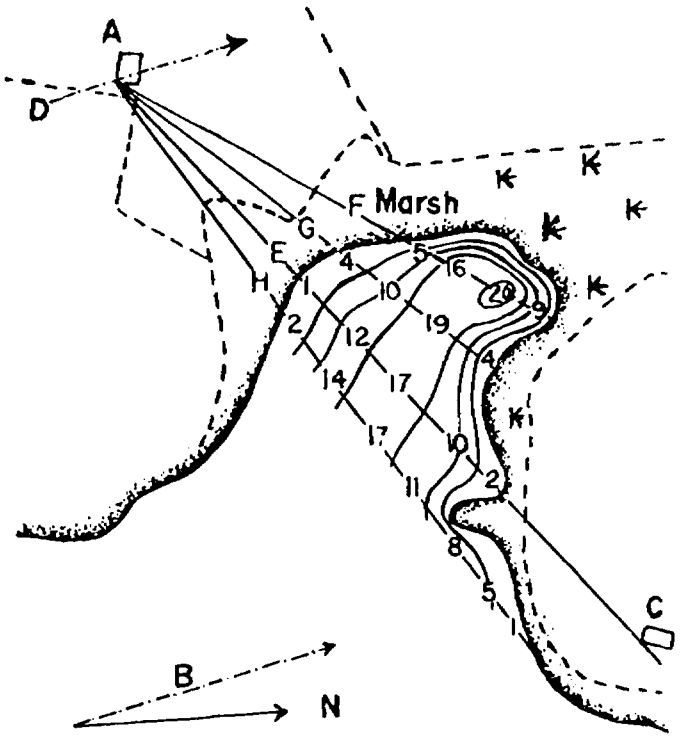


FIG. 9. CONTOUR LINES

WORK SHEET FOR RECORDING SOUNDINGS

Date:_____

Time:_____

Wind Direction :_____

Wind Velocity :_____

Tide Gauge Reading:_____

Profile Line No.	1	2	3	4	5
Direction of Travel					
Sounding No. 1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					

Figure 10

boat. Have the students take soundings at points spaced evenly along each profile line. This can be accomplished by measuring the depth of the water at regular intervals (every 20-30 seconds) while operating the boat at a constant speed (about 1 MPH). Use the landmarks to keep the boat on course. The soundings are made with a lead line; that is, a 4 meter nylon line, calibrated in decimeters, with a two-pound lead weight attached. Keeping hold of the unweighted end of the line with one hand, the student throws the weighted end out ahead of the bow. The line will straighten to the vertical as the boat comes alongside. When it does, the student grasps the line at the water level with his free hand. Keeping the place marked, he then draws the line aboard and reads off the depth, which another student records on a work sheet. This process is repeated on the instructions of the time keeper until the boat has travelled the length of the profile line. Class members should trade jobs after a profile line has been completed so that everyone has a chance to take soundings.

4. Return to the laboratory. Normalize all depths to zero on the tide gauge. For example, if soundings were made when the gauge read +1 decimeter, subtract one decimeter from all measurements. Have each students record the corrected depths at equal intervals along each profile line (Figure 8).

5. The students can now draw contour lines by connecting all of the soundings that are of the same depth (Figure 9). Note that it is most convenient to choose an appropriate contour interval, say 5 decimeters, and to draw contour lines only at that interval. The topography can be emphasized by shading or coloring the areas of equal depth.

Note: If it becomes necessary to save time during this project, the range finder can be used to measure distances both over land and water.

SALINITY DETERMINATION[1]

There is no question that the most distinctive feature of seawater is its saltiness, but to describe it exactly is another matter. The average person is content to know that seawater is too salty to drink, but for anyone concerned with the marine environment it is necessary to use numbers. For many purposes - such as marine engineering - it is enough to know that seawater is a salt solution of about 3.5 per cent, or as oceanographers put it, 35 parts per thousand ($^{o}/oo$). Actually, 90 per cent of all the water in the ocean is within one part per thousand of that mean value, although the total range goes from about $8^{o}/oo$ in areas of extreme precipitation like the Baltic Sea to about $40^{o}/oo$ in high evaporation basins like the Mediterranean and Red Seas.

The marine scientist needs to know salinity very precisely, at least to two decimal places, i.e., $34.73^{o}/oo$, and it is necessary to go to three decimal places, i.e., $34.726^{o}/oo$, to be able to discern the small but significant differences that occur in the deep water.

To measure salinity it is necessary to know just what it is, and that introduces a complication. Salinity is not a fundamental quantity like temperature, rooted in basic scientific principles. Instead, the "salinity" of a seawater sample is a measure of the total amount of dissolved material in the sample, and every element known to occur naturally on the earth has been found in solution in ocean water. Strictly speaking, then, to measure what we call "salinity" you should analyze a water sample dozens of times, to determine the precise quantity of every element present. This would of course be tedious and would require impossibly large water samples.

[1]Most of the material on pp. 27-29 has been adapted from: Redwood Wright, in press. "The Physical Properties of Sea Water." In: Oceanography for High Schools. Available from the Oceanographic Education Center in 1970.

Fortunately, this problem can be met by making use of a very convenient fact: on the few occasions when thorough analysis has been made, the major constituents of seawater were found to occur in nearly the same proportion to each other. In other words, the differences in salinity in seawater are <u>not</u> caused by changing the relative abundance of any of the dissolved materials, but by adding or removing H_2O. The major changes in salinity occur because of rain and snow, which add fresh water, or evaporation and freezing, which remove it.

What this means for the oceanographer is that once the relative proportions of the dissolved substances has been determined, it is only necessary to measure one of them to know them all. One method of determining salinity is to run an analysis for chlorine by titration with silver nitrate, using potassium chromate as an indicator. The chlorinity in °/oo is calculated from the following expression:

$$\text{Chlorinity } {}^{\circ}/_{oo} = \frac{(\text{Molarity of silver nitrate})\,(\text{ml silver nitrate})}{(\text{ml seawater sample})} \times 35.46$$

where 35.46 equals the atomic weight of chlorine. The empirical relationship between chlorinity and salinity, based on the rule of constant proportions, is

$$\text{Salinity } {}^{\circ}/_{oo} = (\text{chlorinity } {}^{\circ}/_{oo} \times 1.805) + 0.03$$

In the titration, the silver ions react preferentially with the chlorine ions to form the white precipitate, silver chloride. At the exact moment when all the chlorine has reacted, the reddish compound, silver chromate, forms. The resulting, permanent color change gives the endpoint of the titration. Although silver bromide and silver iodide are precipitated together with the silver chloride, in the calculations it is assumed that the bromine and iodine had been replaced by chlorine. The titration gives salinity accurately to within about 0.02°/oo.

The accuracy and precision with which salinity can be determined by this method depends on our ability to weigh solids and measure liquids accurately. This is hard to do aboard ship when the weather is rough,

as it often is, and recently a new technique has been developed which is both easier to do at sea and more precise. Instead of chemical analysis, an electronic determination is made of the conductivity of a sample of seawater. Since electrical conductivity depends both on the total amount of dissolved solids, or the salinity, and the temperature, differences in conductivity will represent differences in salinity if the measurements are corrected to a constant temperature.

The salinity and the temperature of a seawater sample determine its density. Salty water is more dense than fresh water because it contains more dissolved matter in a given volume. Similarly, warm water is less dense than cold water because water shrinks on cooling. So cold, salty water will tend to sink and fresh warmer water will tend to rise.

Since the estuary is supplied with fresh water from one source and ocean water from another, layers of water having characteristic densities may form, especially if the wind hasn't been blowing for several days. The importance of layering and the establishment of the thermocline is discussed in the section on plankton (see p. 8).

Since density, temperature, and salinity are so closely related, it is possible to determine salinity from density and temperature measurements with a hydrometer and a thermometer. This procedure, described on p. 44, provides a convenient method of checking the results of the titration with silver nitrate.

Another way to measure salinity is by evaporating a known volume of seawater to dryness and weighing the residue. This method is not dependable because some of the dissolved solids, including chlorides, are volatilized in the last stages of drying.

[illegible] salinity is of prime importance to marine organisms. The body covering is designed to protect the insides of an organism from its external environment. If this covering is calcified like a shellfish's or chitinized like a crustacean's, then the animal is well protected. However, fishes and many other marine organisms have a relatively thin skin. If a fish is placed in saltier water than it has been accustomed to, water will tend to leave the fish's body and the fish will shrink or dehydrate.

Vice versa, if it were placed in dilute seawater or fresh water, water would tend to penetrate inside and the fish would swell or become "water-logged." This phenomenon is an example of osmosis. Life in the aquatic world must maintain its characteristic bodily concentrations of water and salts to survive.

In the group of thin-skinned organisms, which are most affected by salinity changes, there are two major types of adaptations which have evolved. Those organisms which have adapted to tolerate a rather wide range of salinity are known as euryhaline forms while the oceanic forms are usually stenohaline, that is, adapted to endure only small changes in salinity. Both types of marine organisms have body fluids similar in salinity to the water they inhabit, that is, they are isotonic to their surroundings. Since stenohaline organisms cannot survive fluctuations in salinities or prolonged periods of decreased salinities, they must remain in the open ocean and cannot invade the coasts or estuaries.

The manner in which euryhaline organisms remain isotonic to fluctuating salinities is varied. When subjected to sea water of lowered salinity, which regularly occurs along a shore or estuary, some of them can absorb water for a few hours without dying. During high tide, these organisms are flushed with saline water, but during low tide, terrestrial runoff adds fresher water which places these organisms under temporary stress. In time of prolonged salinity stresses, many organisms may close their shells, burrow into the bottom, or migrate to slightly deeper water to escape. Other euryhaline organisms adjust to wide salinity ranges by passively changing body fluids. The American oyster is a good example of this type of euryhaline species since it can adjust to a salinity range of 3-35°/oo, according to Pearse and Gunter (1957).

The pattern of the distribution of species suggests that the highly saline waters of the open ocean are optimum for life. Here a vast number of different species, each adapted to a specific ecological niche, are found. There is a progressive weeding out of stenohaline marine organisms as salinities decline. Few fresh water species may invade the brackish estuarine waters; thus, estuaries are predominately marine. Although there are only a few different estuarine species, their numbers are high.

Some facts of interest prepared from Pearse and Gunter's (1957) treatise on salinity are:

1. An increase in salinity increases the density of seawater.

2. An increase in salinity lowers the freezing point of water (at 35.5°/oo the freezing point = -1.96°C).

3. An increase in salinity decreases the temperature of maximum density of seawater (at 0°/oo, the maximum density is at 4°; at 35°/oo, the maximum is at -3.8°C).

4. The average total surface salinity is 35°/oo, but it varies slightly in the various oceans (in the Mid-Atlantic, the average surface salinity = 37°/oo).

5. Low salinity occurs in polar seas and next to land where fresh water drains into the sea.

6. Salts in the ocean are a result of leaching and land drainage in addition to eruptions of magma through the ages and the original salt in the primeval oceans.

7. Average surface salinity varies with latitude (the salinity is at a minimum at the equator, increases to a maximum at 20° N and 20° S, and then decreases toward high latitudes north and south).

8. The salinity of deep and bottom waters varies within narrow limits (34.6°/oo - 35.0°/oo).

9. Many young marine animals seem to prefer low salinities, then as they mature, migrate to higher salinities.

References

Articles:

Beadle, L. C., 1931. The effect of salinity changes on the water content and respiration of marine invertebrates. Journal of Experimental Biology, Vol. 8, No. 3, 211-227.

Cole, W. H., 1940. The composition of fluids and sera of some marine animals and of the seawater in which they live. Journal of General Physiology, Vol. 23, 575-584.

Metcalf, M. M., 1930. Salinity and size. Science, Vol. 72, 526-527.

Pearse, A. S., and G. Gunter, 1957. Salinity. In: Hedgpeth, J. W., editor. Treatise on Marine Ecology and Paleoecology. Vol. 1, Ecology. Memoir 67, Geological Society of America, pp. 129-157.

Books:

Harvey, H. W., 1966. The Chemistry and Fertility of Sea Waters. London: Cambridge University Press. 240 pp.

Krogh, A., 1939. Osmotic Regulation in Aquatic Animals. London: Cambridge University Press. 242 pp.

Riley, J. P., and G. Skirrow, editors, 1965. Chemical Oceanography. 2 vols. London: Academic Press. 712 and 508 pp.

Sverdrup, H. U., M. W. Johnson, and R. H. Fleming, 1942. The Oceans: Their Physics, Chemistry and General Biology. Englewood Cliffs, N. J.: Prentice-Hall, Inc. 1087 pp.

Lesson Plan

8:00[2] Orientation, attendance, notices.

8:10[3] Lecture on salinity; what is it; why measure it? Discuss ways of measuring it: (1) by taste, (2) by evaporation, (3) by density, and (4) by chemical determination.
Discuss chemistry of titration.
Demonstrate proper technique by doing a sample titration.

10:00 Pick up gear from the lab and storage barn and proceed to the field. In the boat, demonstrate the method of taking samples with the Coke® bottle sampler. Discuss the relationship among salinity, density, and temperature and their effects on the positions of water masses. Half of the group takes samples on a station at the salt water end of the estuary and the other half on a station at the fresh water end.

11:00 In the laboratory, students perform their titrations and calculate salinity.
Discuss results with respect to the physical and chemical properties of seawater and the effects of salinity on the biological population.
If there is time, determine salinity by the density method to check results.

12:00 Lunch - students bring their own.

12:40 Bus arrives to take students back to Intermediate School.

[2]See p. 3.

[3]See p. 3.

Salinity Determination by Titration

Procedure

A. Equipment

1. Ten burettes, 25 or 50 ml
2. Five burette stands
3. Ten Erlenmeyer flasks, 250 ml
4. Ten pipettes, 5 ml, graduated
5. Ten pipettes, 5 ml, volumetric
6. Twenty plastic graduated cylinders, 250 ml, or similar containers for holding pipettes
7. Pipette bulbs
8. Wash bottles
9. Beakers, 250 ml
10. Three reagent bottles: one 1 liter brown glass, for storing silver nitrate solution; two 500 ml clear glass, for storing potassium chromate solution and standard sodium chloride solution
11. Spiral notebook; to be used for permenant records
12. Coke-bottle sampler (Figure 11)
13. Protected thermometer graduated in one-tenth degree intervals and having a range from -10°C to 100°C
14. Ten writing boards or clipboards
15. Worksheet (Figure 12); a copy for each student
16. Ten sample storage bottles, capped and numbered, 500 ml
17. Rowboat or motor boat
18. Life jackets
19. Field tote boxes

B. Reagents

1. Potassium chromate solution (K_2CrO_4), 0.025 molar.

2. Silver nitrate solution ($AgNO_3$), 0.1 molar.
3. Standard sodium chliride solution (NaCl), 0.1 normal.

C. Method

1. Assemble all field equipment (items 12-19) and, taking half the students in the boat at a time, collect samples on each of the two predetermined stations. Samples should be taken with the Coke-bottle sampler to provide a profile from the bottom to the surface. Insert the rubber stopper and lower the sampler to the desired depth. Let it remain there for a minute or so till it comes to temperature equilibrium with the surrounding water. Withdraw the cork by jerking up shaprly on the line attached to it. When the bottle is full (when bubbles no longer come to the surface), retrieve the sample and measure the temperature by the method described on p. 51. Record the temperature on the worksheet. Transfer the sample to the numbered storage bottle and record the bottle number and the depth on the worksheet. Continue sampling at different depths until each student has collected one sample. The surface sample can be drained directly into one of the storage bottles.

2. Return to the lab and determine salinity by titration with silver nitrate.

   a. Fill a 50 ml burette with 0.1 molar $AgNO_3$ that has been standardized according to the procedure outlined below. Make sure that the burette is completely filled below the stop cock and that it is free from bubbles.

   b. With a 5 ml volumetric pipette, place 5 ml of seawater sample in a 250 ml Erlenmeyer flask. (Make certain that students use the pipette bulb and not their mouths to draw solutions into the pipettes.) Enter volume on the worksheet.

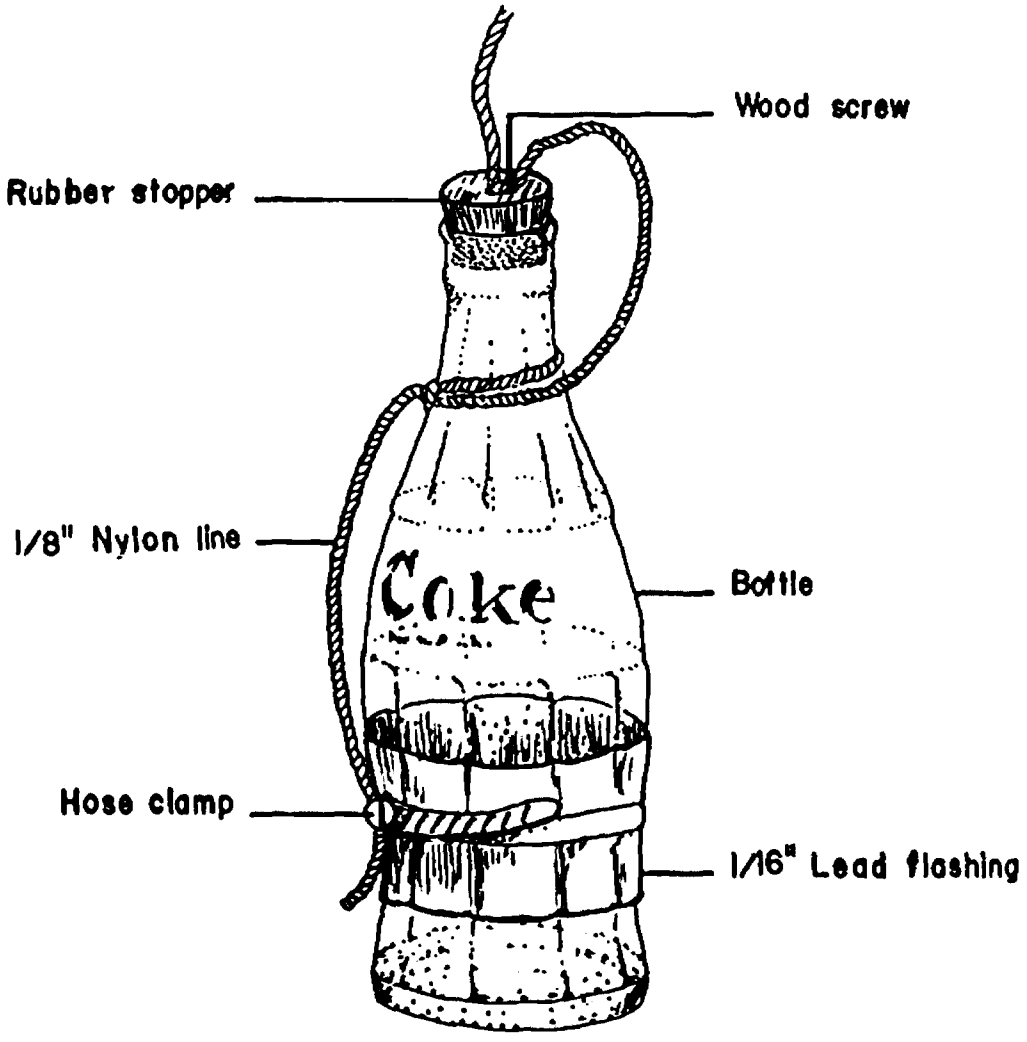


FIG. 11. COKE®-BOTTLE SAMPLER

SALINITY WORKSHEET

Weather Data _____

Name _____ Sea State _____

Date _____ Secchi Disk Reading _____

Sta. No.	Time sample collected	Bottle No.	Temp. of sample	Air temp.	Volume of H_2O titrated	Volume of $AgNO_3$	Molarity of $AgNO_3$	Salinity	Depth of sample

CALCULATIONS #1

Reading of the burette, stop

start _____

CALCULATIONS #2

Reading of the burette, stop

start _____

Figure 12

c. Using a 5 ml graduated pipette, add one ml 0.025 molar K_2CrO_4. Wash down the sides of the flask with distilled water from the wash bottle.

d. Read the ubrette and record the starting level on the worksheet.

e. Using the left hand to operate the stopcock, start letting the $AgNO_3$ solution into the Erlenmeyer flask one drop at a time. Swirl the flask as the titration proceeds to minimize local formation of the Ag_2CrO_4. Wash down the sides of the flask occassionally. The end point has been reached when the solution in the flask turns reddish and remains reddish without further addition of $AgNO_3$.

f. Read the burette and record the stopping level on the worksheet.

g. Repeat the titration till results check.

h. Calculate salinity using the following equations and record the results on the worksheet.

$$\text{Chlorinity (in } {}^{o}/_{oo}) = \frac{(\text{Molarity of } AgNO_3)\ (\text{ml } AgNO_3 \text{ used in titration})}{(\text{ml seawater sample})} \times 35.46$$

where 35.46 = atomic weight of chlorine.

Salinity (in $^{o}/_{oo}$) = (chlorinity x 1.805) + 0.03

i. When he has completed and checked his work, each student should enter a permanent record of his results in the spiral notebook using the form illustrated in Figure 13. To give a clear comparison of the salinity profiles, plot the data from each station on one sheet of graph paper.

PERMANENT RECORD OF SALINITY

Sample Page From Spiral Notebook

Date: _____ Tide: Ebb _____

Time: _____ Flow _____

STATION NO.

Wind Direction: _____ Wind velocity: _____

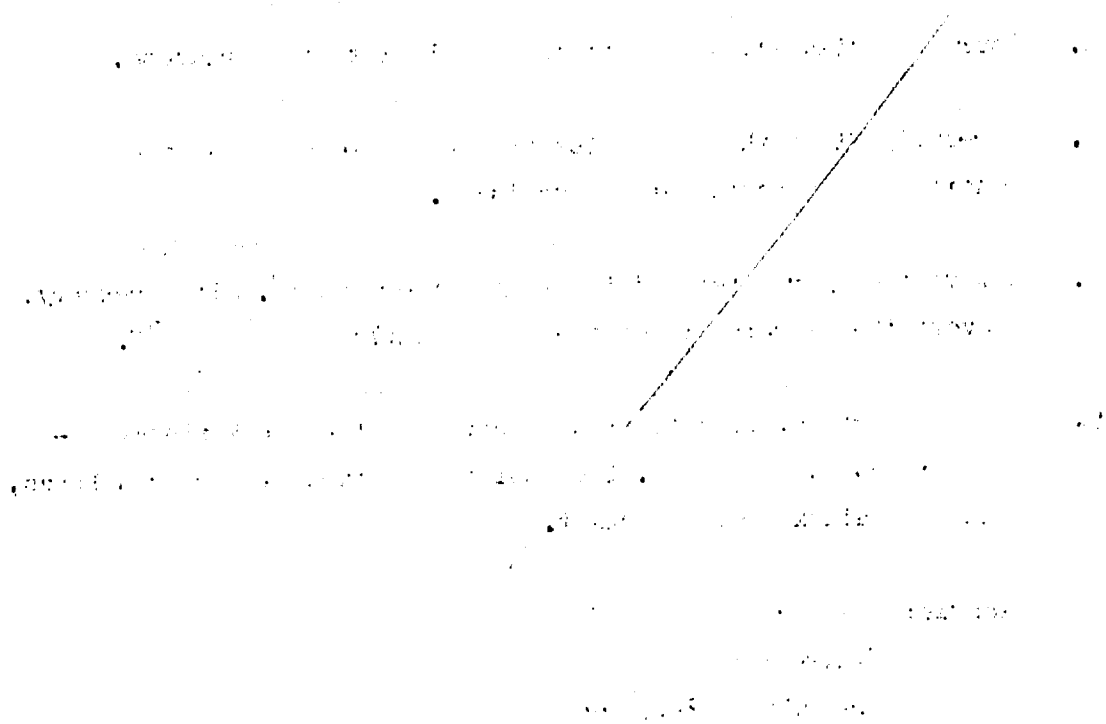
Air Temperature: _____ Barometer Reading: _____

Depth	H_2O Temp.	Bottle No.	Salinity

Figure 13

The end point is marked by the first permanent appearance of a red-brown precipitate of silver chromate. Calculate the exact molarity of the $AgNO_3$ by:

$$\frac{(M\ NaCl) \times (ml\ NaCl)}{ml\ AgNO_3} = M\ AgNO_3$$



Salinity Determination by Density

Procedure

A. Equipment

1. Graduated cylinder, 500 ml; or similar container
2. Hydrometer with range of 1.000 to 1.028
3. Thermometer (the one used in the previous activity will serve)
4. Temperature, density, salinity graph (Figure 14)

B. Method

1. Use samples collected for the titration.

2. Pour approximately 400 ml of the sample into the graduate.

3. Carefully float the hydrometer in the sample and read the density at the bottom of the meniscus.

4. Remove the hydrometer and insert the thermometer. If necessary, convert the temperature of the water sample from ^{o}C to ^{o}F.

5. Using the graph, find the intersection of the density and temperature coordinates and, interpolating between the curved lines, read the salinity at the right.

Example: Temperature = $70^{o}F$
Density = 1.015
Salinity = $22.5^{o}/oo$

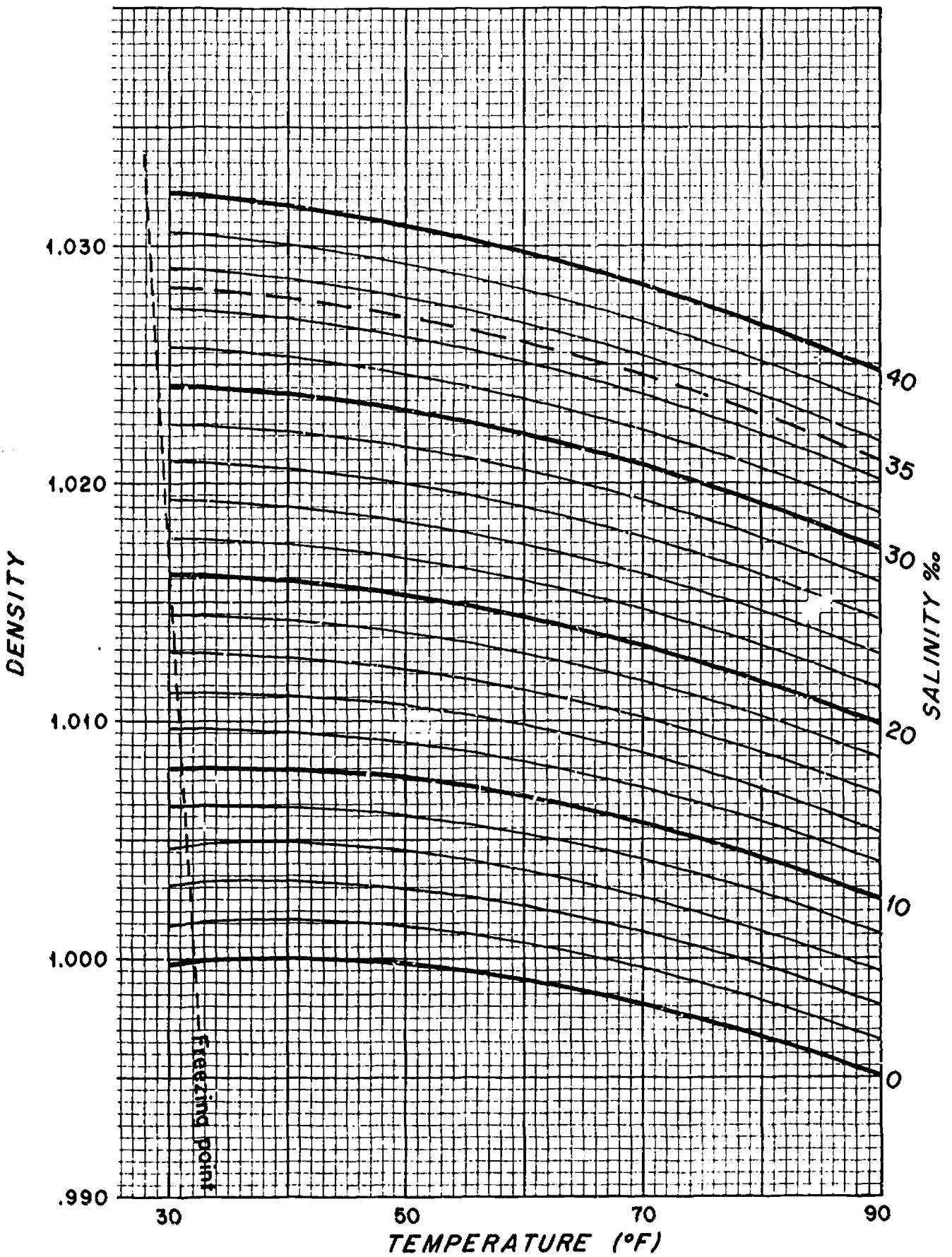

1.030
1.020
1.010
1.000
.990
DENSITY
freezing point
30
50
70
90
TEMPERATURE (°F)
40
35
30
20
10
0
SALINITY ‰

FIG. 14

ENVIRONMENTAL CONDITIONS

Introduction

The students should become aware that certain physical conditions of the atmosphere and the water interact in ways that affect their experiments, and that atmospheric conditions have a variety of effects, both direct and indirect, on the biological population of the estuary. Examples will be noted briefly here; some of them are discussed more fully in other sections of the manual.

Winds and currents affect the accuracy of soundings taken during the bathymetric study because they tend to force the boat off course. Winds also directly affect the height of the tide. A strong onshore wind will pile the water up in the estuary, giving an abnormally high reading on the tide gauge. A strong offshore breeze will produce the opposite effect. Winds and currents have a direct effect on the distribution of the plankton, since these organisms are forced to drift with the water.

One of the indirect effects of atmospheric conditions on the biological population derives from the fact that all organisms require oxygen in order to live. The amount of oxygen dissolved in the water depends largely on the mixing brought about by the winds and upon the temperature of the water as determined by the temperature of the atmosphere. Winds, tides, and rainfall regulate the salinity of the water in the estuary and, therefore, have another important influence on the distribution of the aquatic plants and animals. The saltiness of the water will determine the types of organisms that live in it. The same factors directly affect the results of the salinity determination. Local topography also has important effects. For instance, Bourne's Pond connects with Vineyard Sound through a narrow opening that restricts the ebb and flow of the tide in the estuary. Furthermore, the opening is more or less silted over from time to time causing an otherwise baffingly irregular tidal pattern in the estuary.

These and other important effects of the environment are discussed at appropriate times while the students are engaged in their regular activities, and daily records are kept of salinity, air temperature, barometric pressure, wind velocity and direction, the rise and fall of the tides, and the clarity of the water. In addition, it may be desirable to keep a record of the direction and velocity of currents. Measuring the salinity is itself one of the major activities of the course, and some of the other measurements are made as integral parts of major activities. The remaining measurements can easily be made at odd moments of the day, or in connection with scheduled activities. The records are kept on wall charts in the laboratory so that the students can easily refer to them whenever necessary.

Temperature

Discussion

Temperature is one of the most variable physical features of our planet. It fluctuates during the day, from day to day, and from season to season. It fluctuates less in the sea than on land. Temperatures vary only slightly during the day at any one place on the surface of the open ocean, usually not more than 0.2 to 0.3°C. Shallow coastal water temperatures fluctuate so much seasonally, however, that yearly averages for oceans are not useful. At Woods Hole, Massachusetts, winter temperatures may fall as low as minus 1.7 degrees centigrade, while summer temperatures may reach as high as 23°C.

The range of temperature in the ocean is also less than it is on the continents. The surface temperature varies with latitude from about -2°C in polar seas to about 30°C in open tropical waters. The temperature of deep ocean water is always low, ranging between 4°C and -1°C.

Temperature is one of the most important factors governing the occurance and behavior of life. Plants have essentially no temperature-regulating mechanisms. Animals, however, belong to one of two groups: those that are warm-blooded (the hemoiotherms) and those that are cold-blooded (the poikilotherms). The warm-blooded animals, the mammals and birds, have sensitive mechanisms that regulate their body temperatures at a precise level. By this means, the warm-blooded animals can maintain the optimum rate of biological activity even though the temperature of the environment fluctuates. However, if the fluctuation is too great or if it lasts for a long time, their body temperatures will be changed and they will die. The invertebrates and the lower vertebrates are cold-blooded. For marine animals, this means that their bodies assume the temperature of the surrounding water and change as it changes. Although certain of the higher fishes, notably the tunas, have crude temperature-regulating mechanisms that can keep them slightly warmer than the water they swim in, the rate of the biological activity of cold-blooded marine animals is essentially determined externally by the temperature of the water.

Bodily processes, such as respiration and digestion, will continue for most marine organisms only when temperatures are between 0°C and 35°C. Cold-blooded marine animals will die from heat at lower temperatures than land animals. Most organisms will recover from a cold spell if it is not prolonged, unless ice crystals form in their tissues and cells . However, littoral organisms (shallow water plants and animals) in northern waters are often killed in great numbers by masses of ice that rasp back and forth across the inshore rocks as the tide advances and retreats.

Most marine organisms require a narrower range of temperature for breeding than for their other bodily processes. The common American oyster, Crassostrea virginica, for example can feed and grow at temperatures both lower and higher than those required for spawning. Most animals begin to breed at a definite temperature or temperature change. If the warm season is long, there may be two spawning peaks. Nelson (1928) said that rate of growth and length of larval life of the bivalves was mostly determined by temperature. The role that temperature plays in controlling the supply of nutrients to phytoplankton is discussed on pages 7 and 8.

The annual migration and seasonal movements of many marine organisms are as regular as the migration of birds. Migration has been shown to be closely linked to temperature and light changes.

In the seas, life is most abundant in the colder parts of the world. Cold-blooded marine organisms grow larger in the northern part of their range. Organisms in colder seas grow more slowly, mature later sexually, and live longer.

The following list of facts concerning temperature was extracted from Gunter (1957) and Sverdrup, Johnson, and Fleming (1942):

1. Protoplasm is destroyed at 100°C and at the temperature at which ice crystals form within it.

2. Higher salinities enable organisms to endure colder temperatures, since salt water freezes at lower temperatures than fresh water.

3. Marine organisms survive heat better in high salinities.

4. Organisms with short life spans undergo great population changes with seasons.

References

Articles:

Allee, W. C., 1923. Studies in marine ecology. IV. The effect of temperature in limiting the geographical range of invertebrates of the Woods Hole littoral. Ecology, Vol. 4, 341-354.

Bullock, T. H., 1955. Compensation for temperature in the metabolism and activity of poikilotherms. Biological Reviews, Vol. 30, No. 3, 311-342.

Butler, P. A., 1953. Oyster growth as affected by latitudinal temperature gradients. Commercial Fisheries Review, Vol. 352, 7-12.

Gunter, G., 1957. Temperature. In: Hedgpeth, J. W., editor. Treatise on Marine Ecology and Paleoecology. Vol. 1, Ecology. Memoir 67, Geological Society of America, pp. 159-184.

Nelson, T. C., 1928. On the distribution of critical temperatures for spawning and ciliary activity in bivalve molluscs. Science, Vol. 67, 220-221.

Scholander, P. F., W. Flagg, V. Walters, and L. Irving, 1953. Climate adaptation in arctic and tropical poikilotherms. Physiological Zoology, Vol. 26, No. 1, 67-92.

Books:

Sverdrup, H. W., M. W. Johnson, and R. H. Fleming, 1942. The Oceans: Their Physics, Chemistry and General Biology. Englewood Cliffs, N. J.: Prentice-Hall, Inc. 1087 pp.

Temperature Profile of the Estuary

Procedure

A. Measurements should be taken at predetermined stations permanently marked by buoys. They should be taken at various depths to provide a profile from bottom to surface. It is convenient to make these measurements while collecting samples for salinity determination.

B. Equipment

1. Protected thermometer graduated in one-tenth degree intervals and having a range from -10°C to 100°C.
2. Coke®-bottle sampler (Figure 11, facing p. 40).

C. Method

1. Collect samples with the Coke®-bottle sampler as described on p. 40.

2. After retrieving the sample, place it in a shaded area of the boat and immediately put the thermometer into the sample.

3. When the mercury column no longer moves, read and record the temperature in degrees centigrade. If a Fahrenheit thermometer is used the conversion of Fahrenheit to centigrade can be calculated using the equation $5/9 (F - 32) = {}^{\circ}C$.

4. Surface samples can be taken with a bucket and the temperature measured as in numbers 2 and 3 above.

Measuring Currents

Procedure

A. Equipment

1. Map of the estuary and the surrounding area
2. Several small floats (small jars, with caps painted orange)
3. Stopwatch or a watch with a sweep second hand
4. Buoys and a boat for work in deep water

B. Method

1. Fill the jars with enough water so that the capped tops float just at the surface of the water.

2. Determine the direction of the current flow by drifting the floats and observing their path. Note: If a strong wind is blowing, the surface water may be flowing in a different direction than the sub-surface water. Check this by filling the jars with enough water so that they will float beneath the surface.

3. Using arrows indicate the direction of the current on the map.

4. Choose a place where the current flows in the straightest line and place a float in the water.

5. Measure the time it takes the float to drift from one predetermined position to another. Note: Stakes can be used to mark positions in shallow water; buoys will be needed in deep water.

6. From the distance travelled and the time, calculate the velocity of the current and record it along with the date, the time, and the precise location. Note: Careful records are important because the flow of the current may be closely related to the tides.

Clarity of the Water

The Secchi Disc Method

A. Equipment

The standard Secchi disc (Figure 15) is a circular metal plate 20 cm in diameter. The upper surface is divided into four equal quadrants. Two of them are painted black and the two in between are painted white. The lower surface is painted black in order to eliminate reflection of light from that side. A graduated rope is attached to the upper surface, and a weight is attached to the lower surface to make the disc sink properly.

B. Method

1. Record the conditions under which the experiment is made. Conditions to be included are the following: whether the sky is clear or cloudy; the position of the sun, or the time of day; whether the water is rough or smooth. Ideally, measurements should be made only on sunny days.

2. In the shade of the boat, lower the Secchi disc into the water by the graduated rope.

3. Looking through a viewing box, such as a face mask, record the depth at which the disc disappears from sight.

4. Lift the disc and record the depth at which it reappears.

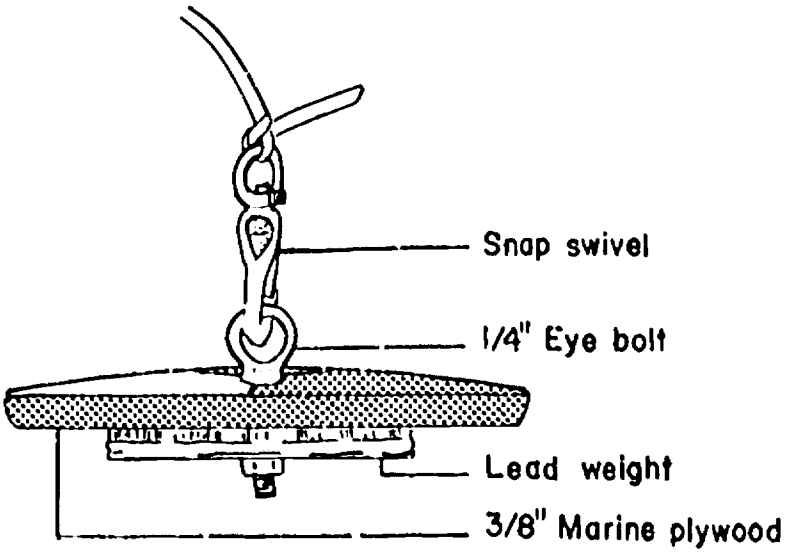
5. The average of the two readings is considered to be the limit of visibility.

6. This information can be gathered most conveniently while the boat is anchored on station for the collection of salinity samples.

Atmospheric Conditions

A barometer and an anemometer are mounted on the laboratory building, and the barometric pressure and the wind velocity and direction should be recorded at the same times each day. The hand anemometer can be used for making necessary measurements in the field.

Measurements of the air temperature and information about the cloud cover are conveniently gathered during the course of the salinity and bathymetry exercises.



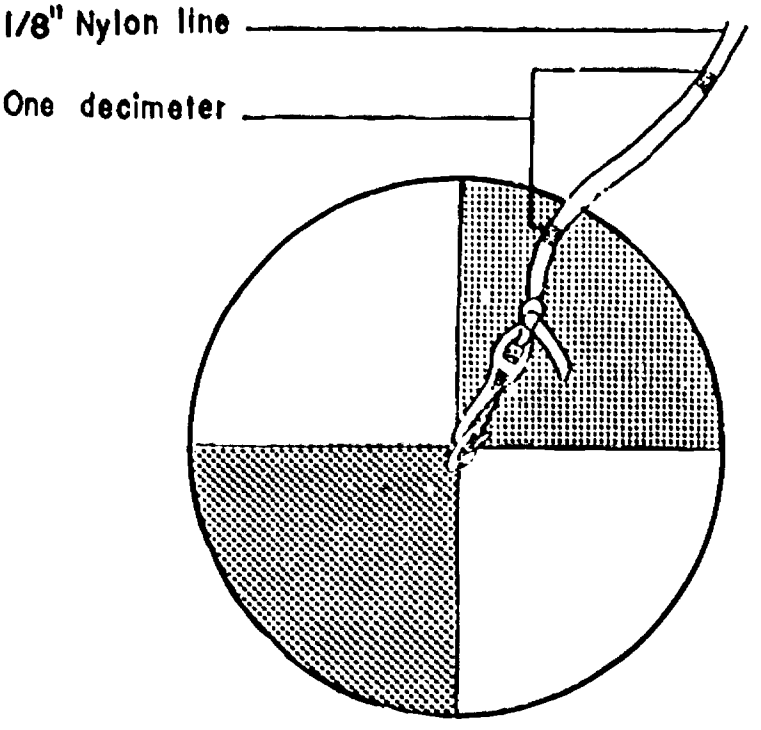


FIG. 15. SECCHI DISC

APPENDIX

A. Materials and Equipment

The following is a list of the supplies needed for the entire course including the amounts necessary for twenty weeks of operation.

Item	Amount
Aquaria	4
Beakers, 250 ml	12
Buoys (made from cinder blocks, rope, and polyurethane floats, or similar materials)	3 or 4
Buckets or wash basins	4
Burettes, 25 or 50 ml	10
Burette stands	5
Coke-bottle sampler	1
Cover slips	1500
Crayons	4 sets
Dip net	1
Dissecting kits	5
Distilled water	5 gal./wk.
Dredge net	1
Eye droppers	12
Erlenmeyer flasks	24
Field tote boxes	4
Fish traps	3
Forceps	12
Formaldehyde, buffered	1 gal.
Glass bottomed viewing box or face mask	1
Graduated cylinder, 500 ml	1

Item	Amount
Graduated cylinders, plastic, 250 ml	24
Lead line	1
Life jackets	10
Magnetic compasses	2
Map of estuary and surrounding area	400 copies
"Marks-a-lots"	6
Meter sticks	2
Metric rulers	6
Microscopes, compound	10
Microscopes, dissecting	5
Microscope slides	1500 (less if you want to reuse them)
Notebook, spiral	6
Nylon line, 1/8"	200 meters
Nylon line, 1/4"	200 meters
Petri dishes	12
Pipettes, 5 ml graduated	12
Pipettes, 5 ml volumetric	12
Pipette bulbs	12
Plankton net	3
Potassium chromate, A.C.S.	1 lb.
Preserving jars, 4 oz.	48
Preserving jars, 8 oz.	48
Protractors	5
Range finder	1
Reagent bottle	
1 l brown glass	2
500 ml clear glass	4
Sample storage bottles, 500 ml	24
Scoop	1
Secchi disc	1
Seine net	1
Sieves	2

Item	Amount
Silver nitrate	4 lb.
Shallow pans	4
Shovel	1
Sodium chloride	1 lb.
Stopwatch or watch with a sweep second hand	1
Thermometers	2
Tide gauge	1
Tripod	1
Vials	1 gross
Wading boots	2 pairs
Wash bottles	12
Worksheets and data forms	400 each
Writing boards or clipboards	10

B. Sources of Supply

1. Plankton net (No. 12 standard silk bolting cloth, 125 meshes/sq. inch, 35 inch net, 9½" ring, #10W 0690)

   Ward's Natural Science Establishment, Inc.
   P. O. Box 1712
   Rochester, New York 14603

2. Dredge net (Nylon net bag, 24' long, steel runners, brass framed ring, #105A50)

   Turtox
   8200 South Hayne Ave.
   Chicago, Illinois

3. Federal Range Finder

   Stoddards'
   50 Temple Place
   Boston, Massachusetts 02111

4. Hand anemometer

   Hartek Instruments, Inc.
   879 West 16th Street
   Newport Beach, California 92660

5. Chemicals, scientific glassware, dissecting microscopes, compound microscopes, dissecting kits, aquaria, burette stands, protected thermometer graduated in 1/10 degree intervals and having a range from $-10^{0}C$ to $100^{0}C$, hydrometer with a range of 1.000 to 1.028, anemometer (wind speed and direction indicator) with dial indoors and outdoor assembly, directional compasses, field tote box

   Nearest scientific supply house

6. Parallel ruler, compasses, protractors, "Marks-a-lots", crayons, notebooks, pencils, clip boards, metric rulers, meter sticks, stopwatch

School supply house or stationary store

7. Wading boots, seine net (approximately, 10' x 4', ¼" mesh), dip net (¼" mesh)

Local hardware store

8. Life jackets

Local marine or aquatic supply store

9. Estuary map - For geological survey maps west of the Mississippi River write to:

Geological Survey, Distribution Section
Federal Center
Denver, Colorado 80225

for maps east of Mississippi River write to:

Geological Survey, Distribution Section
Washington, D. C. 20242

or contact local Town Hall for town survey maps.

10. Tide gauge, tripod, measuring line, seives, Coke-bottle sampler, lead line, Secchi disc

Directions for construction included in this pamphlet