THE SOURCE BOOK OF MARINE SCIENCES

Florida Oceanographic Society
1212 Riverside Drive
Stuart, Florida 33494

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FLORIDA OCEANOGRAPHIC SOCIETY, INC.
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DEDICATION

This Book is dedicated to the memory of John Beakley who was, to the last, the "Grand Old Man of the Sea" for all who knew him. He inspired staff and students and hated the bureaucrats who kept his money thin. He was a teacher, author and originator of much that we see herein. He worked in the sea; he taught about the sea, and maybe, through this Book, he lives in the sea still.

ACKNOWLEDGEMENT

The Florida Oceanographic Society was only two years old when the Florida Department of Education and the Society hosted five young Marine Science Educators to write the first *Source Book of Marine Sciences 1967.* Each had been attempting to teach the Florida marine environment with homemade curriculum, materials and resources. Jack Hopper, James A. Moore and the late Bob Binger of the Department of Education, found the money for a first issue of 5,000. Later a 1970 edition was improved and reprinted as an issue of 10,000 (now mostly unavailable).

The Board of Directors and staff of the Society, realizing that the Source Book needed to be updated and reissued, searched for funding and leadership through the Florida Department of Education, Florida Sea Grant and others. All were interested but could not take on the task. The Board of Directors did, therefore, borrow the funds to get the authors together, have the manuscript edited and finally, the *Source Book of Marine Sciences 1980* is printed.

Among the many who contributed their time and effort to this project, there are those who deserve a special note of thanks.

Frank Pittman of the Environmental Studies Center Jensen Beach, who headed up the writing team and reviewed much of the author's raw material; Dennis Clark, also a teacher at the Center, for his assistance to Frank in both coordination and review; all of the authors, who shared their own experiences to make this publication possible; the original authors, Renwick, Taylor, Golden and Beakley; Mark Perry, Executive Secretary for the Society and much more; C.W. "Pete" Huff, former Board member and financial back-stop for the project; Dr. Sheldon Dobkin, marine biologist at Florida Atlantic University, who was the very first acadamecian to support the upwelling of interest in marine science at the secondary school level. He also edited the new Source Book along with Bill Hammond, Environmental Education of Lee County Public Schools, Frank Kapp, Office of Environmental Education, Florida Department of Education and Will Hon, Environmental Specialist, Skidaway Island, Georgia who also contributed magnificent illustrations of water birds and other biological illustrations.

And more: Bob Bergen and Marge Gordon of Pine Jog Environmental Center, West Palm Beach, who reviewed much of the authors' raw manuscript material; then there are Richard Granfield and Linda Wasenius, his secretary, who typed and copied the drafts in the office of Dick's Architectural firm; The Society's Board of Directors, Cliff Perry, J. Alex Raphel, Rev. Arthur Smith, James Littman as original founders, and new Directors, William Turknett, Stanley Huddleston, Richard Granfield, Glenn Massnick Jr., Lee Harris, and Thomas Reiling; Pat Lyons and Evelyn Harris, secretaries who helped type the many rough drafts and revisions; and of course all those whose names may have inadvertently been left out.

Finally, credit should go to the thousands of high school students who waded and seined; dug and measured; froze and blistered; loved and cared for the creatures, critters, plants and sea scapes . . . learning all the way. Let us hope that the 1980 edition will continue these grand adventures in learning.

As for me — I walked with and among those who worked hard to teach the children and who will make the *Source Book of Marine Sciences* live for the children to come. We owe them all — thank you.

Sincerely,

E. Ray Roberts

INTRODUCTION

This Source Book was prepared following the same format as the original Source Book of 1967-68 and with the same intention. The materials contained in this Source Book were developed with the primary aim of assisting science teachers at all levels and subject fields to improve their curriculum by making better use of the local environments.

The Source Book is intended primarily for secondary school levels but it can apply as a supplement to several levels of marine science education. Each unit was developed as an individual concept and therefore may be taken in any order of learning preference. There is no teacher's supplement for the Source Book. In each unit there is an "Introduction" or "To the Teacher" explaining the overall concept or methodology to be used. "To the Student" introduces the student to the concept and begins the student's activity in the unit. After the activity "questions for consideration," "graphic analysis" or other recommendations serve to follow up or reinforce the exercise. Also at the end of each exercise there is a list of suggested "references" for a more in-depth study of the particular subject area.

Most of the units present "hands on" activities both in the lab and in the field. Other units introduce a concept which may be expanded by the teacher according to the local curriculum. The Source Book 1980 stresses the consideration of the overall marine environment as the student learns to analyze a system or environmental "niche" rather than one specific parameter or organism. "Introduction to the Analysis of Sea Water," "Biochemical Survey of an Aquatic Ecosystem" and "Introduction to the Mangrove Ecosystem" all employ the environmental concept. Environmental education continues to receive increasing attention not only in Florida but throughout the United States and marine science is perhaps the most appropriate and effective tool for learning about the total environment.

The Source Book of Marine Sciences is intended for use wherever any one concept may be applied. The original volumes were requested from a variety of areas such as the Phillipines, Australia, South America, Alaska and New England. As the Book was authored in Florida, it may seem to present many viewpoints applicable to the environments of this state. However, during the development of the Source Book, efforts were made to expand it's direct usability to the coastal, southeastern United States.

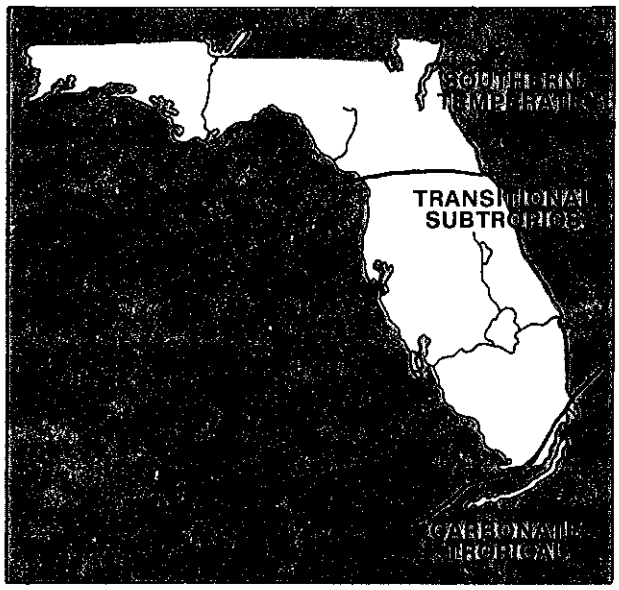
In the near future there should be attempts to coordinate marine science educational materials and we do hope they will be successful. The Source Book of Marine Sciences, we believe, is now and will continue to be a useful part of these materials in the years to come. In the overall interest of developing additional resource materials for marine science and improving this Source Book, teachers are encouraged to forward their reactions, recommendations and their own locally developed lab and field exercises to Florida Oceanographic Society, 1212 Riverside Drive, Stuart, Florida 33494.

CONTENTS

INTRODUCTION TO COASTAL ECOLOGY

Florida's coastal systems respond, primarily, to changes in latitude, to differences in substrate (usually sand, mud or rock) and in varying degrees to differences in fresh water inputs, coastal current systems, human intervention, and to natural catastrophes such as hurricanes or severe cold snaps. The coastal systems of Northern Florida are very similar to those of Georgia and the Carolinas, and to those of the other Gulf coast states (Alabama, Mississippi, Louisiana and Texas) with a preponderance of identical or similar species. The coastal systems of extreme South Florida are unique in the continental United States, in that they include coral reefs of the tropics.

Although these systems look very different they all respond to the same basic rules for existence of living systems, and they have many common characteristics.



COASTAL ZONES OF FLORIDA

THE CARBONATE TROPICAL SYSTEM

The Florida Keys and, to a lesser extent, the lower East Coast and Florida Bay regions have a common denominator. The substrates of the area are either limestone rock or limestone sands and muds, locally termed "marl." The limestone rock comes from two main sources, coral reefs and oölitic limestone deposits, hardened into a soft crumbly rock known as Miami Oölite. The upper Keys from Soldier's Key in Biscayne Bay to the Newfound Harbor Keys at Big Pine are formed from ancient coral reefs. A good cross-section of this 120,000 year old coral reef can be seen in the Cross Key Canal at Key Largo.

The lower keys from Big Pine Key to Key West, the floor of Florida Bay, and the floor of most of the Everglades region is formed of Miami Oölite. This oölitic limestone was laid down about the same time the Upper Keys were living reefs, and is composed of a mixture of fossilized bryozoans, coralline algae and oöids which are tiny spheres of calcium carbonate precipitated from warm shallow salt water onto even tinier nuclei. This precipitation process occurs every summer in Florida Bay and on the Bahama Banks.

The upland living marine related systems of the Keys are characterized by tropical hardwood hammocks and stands of slash pine on the higher ground; mangrove swamps and sea grass beds in intertidal and shallow water zones, and living coral reefs in deeper water.

All of Florida's coastal beaches are exposed to wave and wind action. Sands of quartz or carbonate is wind blown inland to form dunes which migrate before the wind until stabilized by plant roots. Waves carry material from sea to beach, leaving it behind in ragged piles as the tide recedes. This "wrack" line of decaying plants and animals becomes the base of yet another food wed, again with crustaceans such as ghost crabs and amphipods acting as intermediaries converting plant to animal protein.

THE TRANSITIONAL SUB-TROPICS

The area from Ten Thousand Islands northward along the Gulf Coast to Levy county (Cedar Key) and from Biscayne Bay northward on the east coast to Volusia County (Daytona Beach) is sub-tropical in species composition most of the time. We will consider red mangrove the indicator species for this zone.

The Gulf Coast is a low-energy coastline, subject to large waves only in hurricanes and other major storms. The substrates are mostly quartz-derived sands and muds, and large expanses of soft-bottomed shallow water are common. The Floridian Plateau extends offshore as a gentle sloping platform a hundred miles or more wide. This large expanse of shallow water makes the region biologically very productive, and also makes it susceptible to inundation by hurricane surges up to 35 feet. Barrier islands are scattered. Beach sands are fine quartz. Red mangrove is the dominant salt marsh species of the southern region of this zone. Northward, cord grass becomes more plentiful and the red mangroves fewer and smaller, finally appearing as isolated bushy small trees in a predominantly cordgrass marsh at the northern edge of the zone. The same gradation of red mangrove to cordgrass occurs on the east coast.

The east coast faces the Atlantic Ocean and typically

presents most of the features of a high energy coastline. A barrier island is usual with a dune system on the ocean-facing side and lagoon system behind the island. The continental shelf is narrow (only a mile or two wide off Palm Beach) and the ocean bottom deepens very rapidly compared to Gulf side. Hurricane surges on this coast typically reach a maximum of 15 feet.

In southern Florida, reefs of coral or coquina are common offshore paralleling the coastline. Generally clear water conditions prevail northward to about Jupiter Inlet most of the year, and in summertime to about Vero Beach. The beach sands grade from nearly pure quartz at Daytona Beach to nearly pure carbonate sands south of Miami.

SOUTHERN TEMPERATE COASTLINE

Along the southern temperate coastline north of Daytona Beach and Cedar Key, mangroves lose their dominant role in the salt marsh system to two grasses: cordgrass, *Spartina alterniflora* and black rush, *Juncus maritima*. The salt marshes of north Florida are largely river-dominated. That is, large components of both their fresh water and nutrient input come from rivers such as the St. Johns and St. Marys on the east coast and the Suwanee, Apalachicola and Yellow Rivers on the Gulf Coast. The continental shelf on both coasts is wide with a gradual slope and is less steep on the Gulf side.

COMMONALITIES AND DIFFERENCES

All of these coastal systems must obey the same "rules" or "laws" of nature, and many similarities can be found among them. The team may have a different name, the players wear different colored uniforms, but the rules of the game are the same.

One of the "similar-differences" is based on the detrital food web found in any salt marsh. The role of mangrove leaves in a tropical marsh is played by cordgrass stems in a temperate marsh; sheet flow fresh water drainage in the Everglades is replaced by riverine fresh water input in many north Florida marshes. In any case, a typical salt marsh system will have three basic types of producers present, each supplying energy to its own food web.

Emergent vegetation such as mangroves and cordgrass supply a detritus-based food web in which crustaceans play a vital intermediary role.

Submerged vegetation in shallow waters outside the marshes provide food for a complex grazer-based food web, a substrate for sessile organisms and a habitat for mobile organisms.

Plankton drifting in the tidal currents throughout the marsh supply food to the planktonic food web (which includes at some time or other the young of almost everything in the marsh) and to the myriads of filter feeders living in, on and under the many substrates and surfaces found in the emergent and submergent vegetation zone.

These three basic energy producers in any salt marsh are supplemented by a fourth: *surplus food* transported into the marsh by sheet flow drainage or by a river system. This may be a major component of the total energy flux of a marsh.

The great productivity of salt marshes leads to the development of deposits of mud, often several feet deep, composed of decayed plants and animals. Decomposition in these soft, sticky muds is mostly anaerobic resulting in the "rotten egg" smells produced by hydrogen sulphide and sulfur dioxide, a characteristic shared by salt marshes around the world. These muds act as a nutrient bank, and an occasional hurricane may stir up the muds, putting the nutrients back into circulation. Tides carry nutrients from marsh to ocean in the form of suspended organic particles, dissolved organic chemicals, and organisms going to sea after feeding in the marsh.

MAN'S INFLUENCE IN THE COASTAL ZONES

Man's influence on the Florida Keys has been great. Building, construction and associated paving on high ground; dredge and fill in intertidal and shallow water zones; destruction of coral reefs by siltation, specimen collecting and boat anchors; and finger canals dug into the solid rock to provide boat access; all are destructive influences.

Man's influence on the coastal systems of the Southern Temperate Coastline and the Transitional Sub-Tropics is most noticeable around cities. Here natural systems have been replaced by concrete shores and oozy bottoms. Tentacles of construction stretch outward from these nuclei for miles along the coastline. Construction in population centers has replaced natural dune systems, salt marshes and lagoons. Pockets of natural areas remain, but even they are effected by disruption of fresh water input (as the Ten Thousand Islands region, and eastern Florida Bay) or by chemical pollution of rivers (as the pulp mills of nothern Florida) and by other activities of man.

INTRODUCTION TO THE MANGROVE ECOSYSTEM

Along the Atlantic and Gulf coasts of Florida lies a band of salt tolerant trees called mangroves. These trees are products of thousands of years of evolution and adaptation to Florida's coastal environment and provide a major marine ecosystem vital to the health and productivity of Florida coastal waters.

Historically, mangroves have been considered as nuisance trees, creating mosquito breeding grounds and inhospitable swampland. Thousands of acres of mangrove forests have fallen to the bulldozer and dredge and fill operations as developers seek to provide their buyers easy access to the ocean as well as an unimpeded view of the water. Hundreds of acres of mangrove fringe and shallow bay bottom have been dredged and filled to create new premium waterfront property which increases the developers profit margin.

Botanists recognize four species of mangroves in Florida.

1. *Rhizophora mangle*—the red mangrove which ranges southward from Levy County on the Gulf Coast to Volusia County on the Atlantic Coast.
2. *Avicennia germinans (nitida)*—The black mangrove ranges throughout the Gulf Coast but only as scattered shrubs north of Levy County and found up to St. Johns County on the Atlantic Coast.
3. *Laguncularia racemosa*—The white mangrove is found northward to Hernando County on the Gulf Coast and up to Brevard County on the Atlantic side.
4. *Conocarpus erecta*—The Buttonwood or Button Mangrove. Usually found with the mangrove but it's range is limited to South Florida.

Ecologists recognize five basic mangrove communities formed by the above mentioned species.

1. Basin forests—Inland depressions of highly varying salinity.
2. Riverine forests—Flood plains of major drainage creeks and rivers.
3. Fringe forests—Mainland and island coasts fronting bays and open seas.
4. Overwash forests—Small islands and land projections periodically overwashed by tides.
5. Dwarf mangrove forests—Mangroves growing in areas with reduced nutrient levels.

In the early 70's the attitude of some Florida citizens and their lawmakers began to change about the importance of mangroves. Scientific investigations were providing increasing amounts of data concluding that mangrove forests were necessary and vital ecosystems to the health and productivity of Florida's coastal waters.

Floridians began to realize that some serious mistakes had been made by allowing their valuable state resource to be destroyed for short term profits at the expense of all Florida citizens. Laws were made to protect at least some of the mangrove forest areas, but to date enforcement of these laws has been subject to inequities because of a combination of political and economic pressure of monied special interests compounded by a lack of education of the general public about the value of mangroves to all of Florida.

Bill Hammond of the Lee County Public School System in his paper "A Case For Mangroves" summarized so very well the role of the mangrove ecosystems to both man and nature.

Based on past and present scientific research, Dr. Hammond presents six major reasons why mangroves must be protected in Florida.

1. *Productivity*—The predominantly red and black mangrove detrital based system is probably one of the most biologically productive in the world. Many marine fishes and invertebrates depend on this system for survival during some stage of their development.

2. *Shoreline Protection and Stabilization*—Red and black mangroves play an important role as back-bay land stabilizers. The red mangroves, with their characteristic prop roots, assist in reducing runoff siltation by acting as silt traps. In addition, they act as wave energy absorbers, thereby preventing wave and tidal erosion along shorelines. They serve as important coastal hurricane buffers, protecting upland populations and properties from flooding and wave-generated destruction by absorbing and diffusing wave shocks and mechanically retarding rising floodwaters.

3. *Runoff and Pollution Filter Trap*—Research at the University of South Florida indicates the mangrove-dominated estuary serves as a very effective absorber of pollutants, particularly raw domestic sewage. Mangroves, and the organisms associated with them absorb and utilize these types of pollutants at no cost to the taxpayer.

4. *Wildlife Habitat Area*—The red and black mangrove forests are major breeding and feeding grounds for a majority of our wading birds, estuarine fish, and invertebrates. In addition the mangrove fringe zones serve as a nursery ground for many of our important food and gamefish.

5. *Mangrove Dollar Economics*—Economist John McQuigg for the 1968 Florida Legislature and later Drs. J.G. Fosselink and R.M. Pope of Louisiana State University and Dr. E.R. Odum of the University of Georgia have estimated the dollar value of estuarine regions in general to

be between 50,000 and 80,000 dollars per acre at 1977 level prices. Mangrove estuaries being the most productive of all estuaries are valued at the higher dollar figure. It is a different concept to understand, but the dollar value of mangrove areas directly influences the cash flow of our state economics and directly or indirectly profits every citizen of our state.

Mangrove areas destroyed for development only benefit the developer. The contribution of developed mangrove land to the county tax rolls is more than absorbed by the need for city and county government to provide services to the new area.

Florida is dependent on a healthy marine environment to support our multi-billion dollar tourist and marine economy. By destroying mangroves we are in effect "biting the hand that feeds us."

6. *Uniqueness of the Mangrove Forests*—It is human nature to cherish what is unique. In all of the continental United States, mangrove forests are unique from Florida around the Gulf through Texas. We have an environment worthy of protecting for all to see and enjoy. We must remember that once an estuary is destroyed, it is forever destroyed. We would be foolish and shortsighted as a state and a nation to allow this remaining valuable resource to be destroyed forever.

INTRODUCTION TO THE ENERGY FLOW IN A MANGROVE MARSH

What makes a mangrove zone productive? Why are estuaries in general productive? We have said much so far about this productivity but offered little in the way of explanation as to how this comes about. In the late 1960's two University of Miami scientists, Dr. Eric J. Heald and Dr. William E. Odum earned their doctorates by uncovering the role of mangrove detritus in the energy flow of Florida's mangrove zone coastal waters.

Their research, done in the New River estuary system of Everglades National Park, found that while small amounts of phytoplankton and benthic algae were eaten, it is the fragmented leaves of the red mangroves which are the primary producers in mangrove-dominated zones.

Dr. Heald discovered that mangrove tree leaf fall results in an annual production of leaf debris exceeding three tons (dry weight) per acre. Their importance to the ecosystem of the estuary starts after they fall from the tree and begin to decompose. The speed of decomposition depends on whether they fall on the land or in the water — water being the more efficient environment for decomposition. Physical breakdown of the leaf is speeded by crabs and amphipods (relatives of shrimp about the size of a grain of rice) whose agile claws shred the decomposing leaf into smaller fragments. Within six months more than 30% of the leaf material has been grazed by scavenging aquatic organisms. Three more months are sufficient for the leaf to be reduced to fragments no larger than 1mm. in width.

During this breakdown process particles of mangrove litter became covered with bacteria and fungi which use the leaf material as food. These organisms, which are themselves rich sources of vitamins and protein increase the caloric content and protein of each leaf particle. Thus, the particle becomes an increasingly valuable food source for larger animals as it decreases in size and becomes more heavily coated with micro-organisms. Heald found that at the end of 12 months the mangrove particles were about 22% protein compared to about 6% when they left the tree in the intact leaf. These leaf particles, now called detritus, get smaller and smaller as diminuitive animals ingest them, devouring the bacteria and fungi and excreting the indigestible cellulose bit. Each excreted bit gets another collection of micro-organisms which go down the gullet of yet another sea creature. The process is repeated again and again. The detritus, the vehicle upon which the bacterial and fungal protein rides, can hardly get too small, for when it reaches less than 0.005mm or 10 thousandths of a millimeter it may combine with other small particles in conglomerates, and the cycle is continued. As much as one half of this rich detrital food is flushed into the sea from estuaries during spring-summer runoff periods delivering a rich food supply to the sea and it's larval plankton forms.

William Odum examined the stomach content of over 6,000 organisms representing approximately 90 species of fish, shellfish, oysters, clams and insect larvae to discover what served as their food in the estuarine environment. From such observations, Odum and Heald were able to reconstruct the biological pathways of the food energy flowing through estuarine community and to assess the relative value of mangrove material to fishes and other members of the animal community in the estuary. (See Energy Flow pages 6 and 7.)

MANGROVE MARSH

TO THE TEACHER

This exercise involves one field trip and one or two periods in the classroom/laboratory. This field trip can be used to collect specimens for several different labs depending on location and season. You can also do "Introduction to the Analysis of Sea Water" (page 25).

TO THE STUDENT

The energy flow, also known as the food chain, food cycle or food web of the estuary or the open sea is so complex that it is not possible to illustrate in any form other than a three-dimensional model. Even this would be further complicated by the need to show seasonal variations. It should be understood that the feeding habits of the animals vary according to season and also individual species vary their diets in different parts of the estuary or ocean.

Many organisms depend on the detritus food chain based on mangroves. Many depend on mangroves for hiding places or a place to attach and live. Others depend on organic sediment formed by detritus as a place to live and feed.

In the North River estuary of the Everglades, detritus accounts for 80-90% of the nutrition of a number of species

of crabs, worms, insect larve, shrimp, and small forage fishes. These detritus eaters are prey for over sixty species of juvenile fishes which live in the mangrove bordered estuary for part of their lives. Among these are the tarpon, snook and ladyfish, which utilize the mangrove belt from the time they reach the estuary as post-larvae. Gray snapper, sheepshead and red drum spend the first few weeks of their lives in the grass beds of Florida and Whitewater Bays, and then move into the mangrove habitat for several years. Other game fishes which are found in and near the mangrove zone are spotted seatrout, jack crevalle, gafftopsail catfish and jewfish. As young fish, they eat amphipods and insect larvae then, as they grow larger, they consume shrimp and crabs. They also consume foragefishes such as the tidewater silver sides, the silver jenny, and the rainwater killifish, which in turn eat detritus feeders.

The value of the sea grass, macro algae and mangrove nursery areas for commercial fin fish species is thus very great. Commercial species of shrimp also depend on estuaries. Even open ocean organisms depend on protein derived from estuaries.

PURPOSE

Examination of mangroves and associated orgamisms *in situ* to understand detritus based food web.

MATERIALS

A. Field
Field guide to common marine organisms
Mangrove identification sheets
Metric ruler
* Shovel
* ¼" hardware cloth sieve
Seine
Assorted collecting containers
* Optional

B. Laboratory
Magnifying glasses and/or dissecting microscopes dishes, watch glasses or specimen dishes.
Shallow sorting pans
Assorted specimen jars

PROCEDURES

A. *In the Field*
1. On reaching chosen site each student is to:
   a. Record time of day and tide conditions.
   b. Identify 4 mangrove species (if all are present).
   c. Determine if and where salt is excreted from red mangroves.
   d. Record species on red mangroves prop root.
   e. Measure length of root encrusted.
   f. Identify mangrove crab, *Aratus pisoni.*
   g. Identify male and female fiddler crabs, *Uca* sp.
   h. Determine if *Uca's* large claw is used in feeding.
   i. Count crab holes in area 0.5m x 0.5m.
   j. Record and count organisms in one handful of red mangrove leaf fall.
   k. Record and count organisms in seine haul.
   l. Examine fish for color patterns
2. As a group each class is to collect one small sample of submerged sediment (baby food jar).
   a. Collect *one* of each organism found.
   b. Collect about 20 cm x 20 cm x 20 cm red mangrove leaf fall.

B. *Back in the Lab*
1. Examine leaf fall under magnification, identify and count organisms.
2. Repeat #1 for sediment sample.
3. Preserve all specimens from field and laboratory, label and store for resource use.
4. Pool all data

GRAPHIC ANALYSIS

1. Graph crab hole numbers and determine average number per meter.
2. Graph numbers of dominant organisms on prop roots and determine average number per meter.

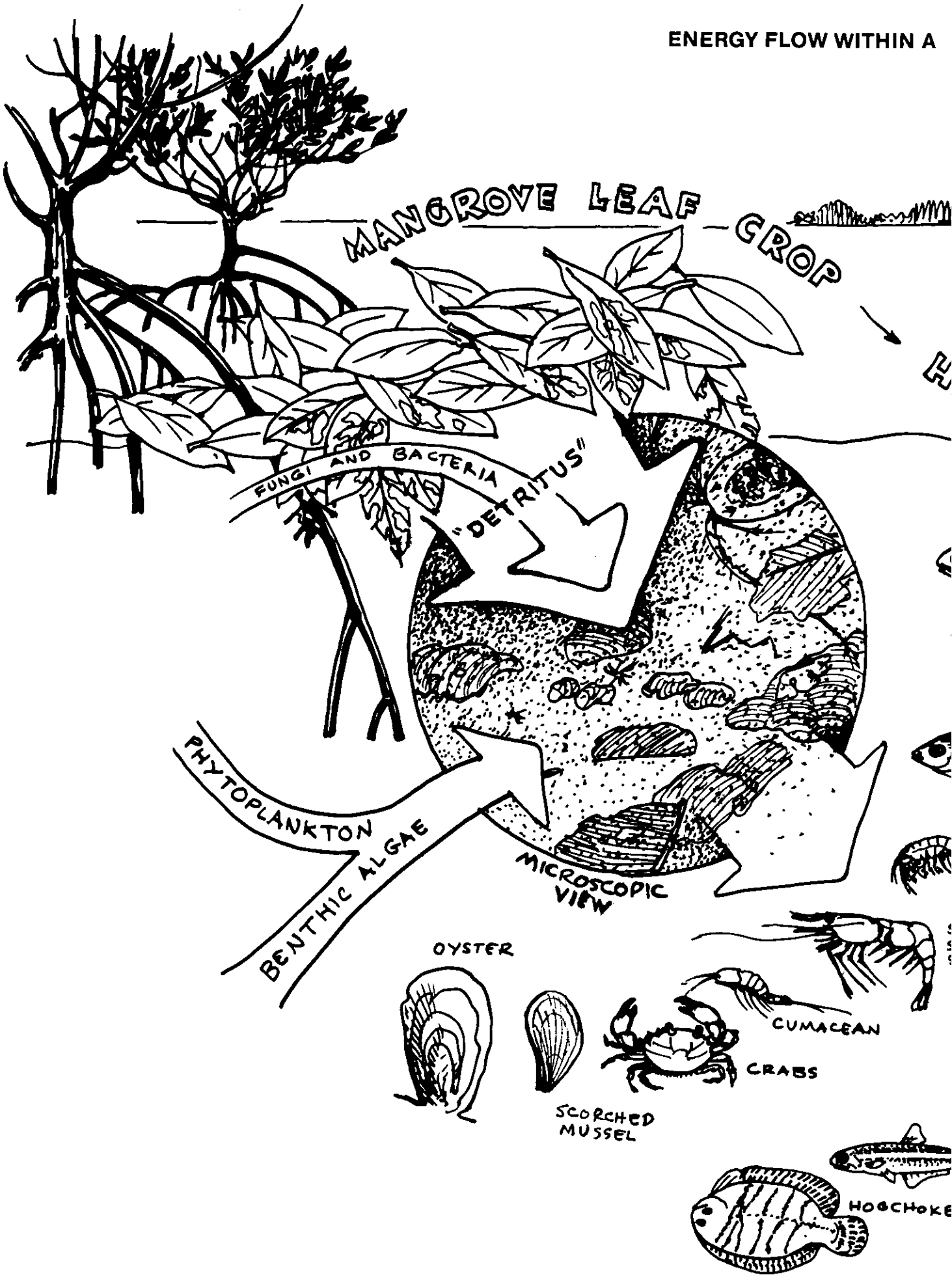
QUESTIONS FOR CONSIDERATION

1. Why do mangroves excrete salt?
2. Why do different mangrove species have different salt excreting capabilities?
3. What does *Uca* do with its large claw?
4. What might *Aratus pisoni* eat? (Not leaves)
5. Why are so many fish dark colored on the back and light colored on the belly?
6. What non-aquatic class of animals is a dominant predator in salt marshes?
7. Why is it dangerous to eat oysters from polluted waters?
8. Why isn't it dangerous to eat mackerel caught in polluted waters?
9. What is the principal food for infauna here? (Infauna are animals living in sediments.)

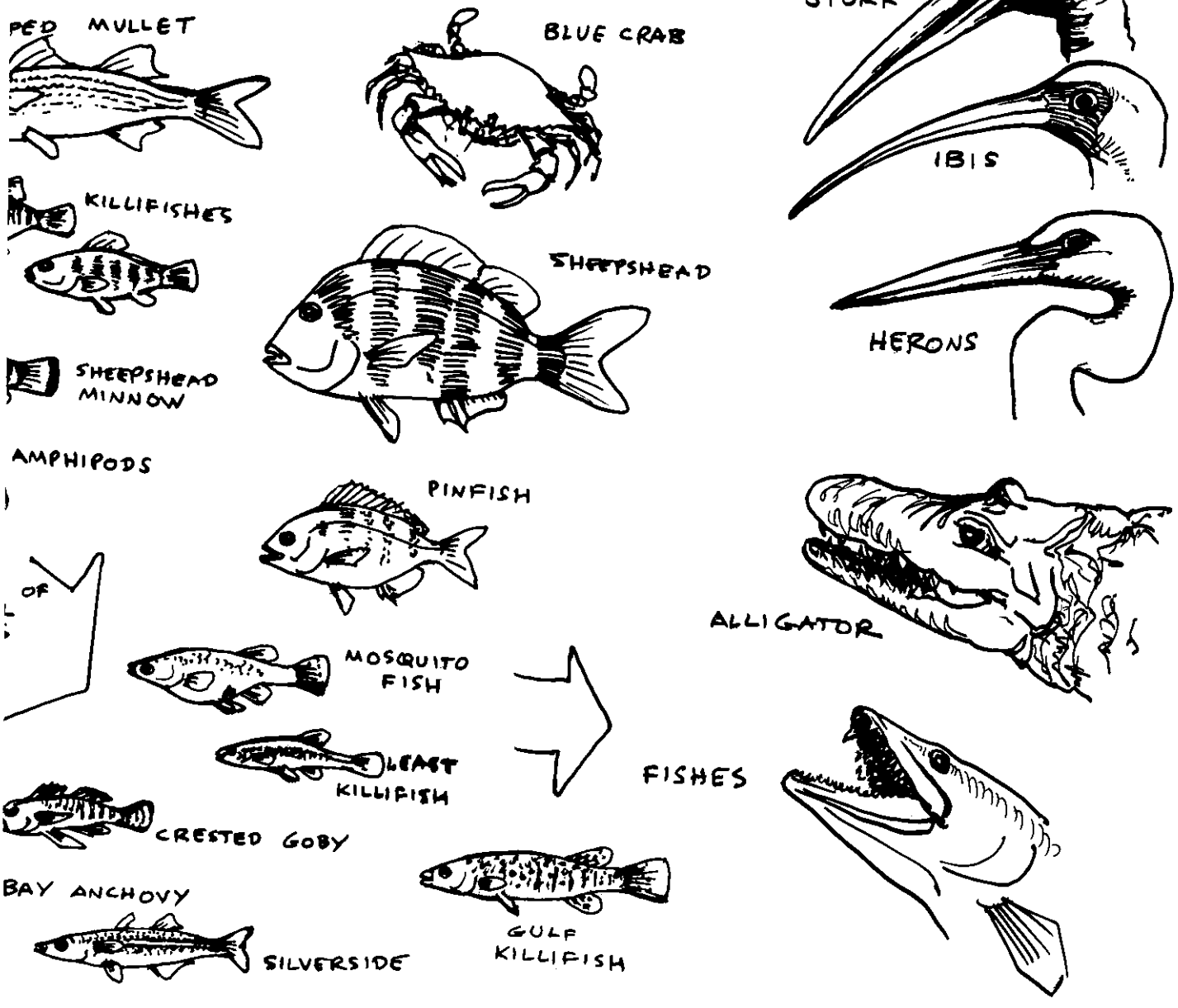
MANGROVE IDENTIFICATION

THE RED MANGROVE—*Rhizophora mangle*

This easy to identify tree is usually found adjacent to the open water fronting bays, rivers, creeks, and open oceans. The characteristic "prop roots" arising from the trunk and branches make the tree easy to identify. The waxy leaf is dark green on the top surface and lighter green on the underside. The leaves are positioned in an opposite arrangement on the stem. The flower is a downward-facing pale yellow which


MANGROVE LEAF
CROP
FUNGI AND BACTERIA
"DETRITUS"
PHYTOPLANKTON
BENTHIC ALGAE
MICROSCOPIC VIEW
OYSTER
SCORCHED MUSSEL
CUMACEAN
CRABS
HOGCHOKE

BIVORES → CARNIVORES
(SEVERAL LEVELS)



produces a 2.5 to 2.0 cm seed. The seed germinates on the parent tree producing a viviparous seedling from 20-30 cm in length. When these seedlings drop from the parent tree they may stick upright and root under the parent tree or be carried by the tides many miles from the parent where they take root in muddy or rocky areas. Mature trees will grow to 25 m tall.

The Red Mangrove tends to deliver its detrital energy flow to the estuarine bays with every high tide, and particularly from high upland runoff during the rainy season.

THE BLACK MANGROVE—*Avicennia germinans*

The Black Mangrove is the most salt tolerant of the mangroves and is often found in depression marshes and riming salt ponds in the interior of coastal islands. These environments produce highly saline conditions when seepage and tidal waters are trapped and evaporate leaving salt behind, the conditions in which only the most tolerant of marine plants can survive. The Black Mangrove leaf will often have visible salt crystals formed on the outer surface of the leaf due to excretion of excessive salt from the plant tissue.

The leaves are smaller (5-10 cm) and more narrow than the Red Mangrove leaf. The leaf is pale shiny green above and lighter "fuzzy" gray-green on the underside. The leaf has an entire margin and is arranged in an opposite fashion on the stems. The flowers are small, white, and yellow at their base. The seed is shaped like a large lima bean and is green in color. The Black Mangrove bears viviparous fruit. Mature seeds fall to the ground after emerging from the seedcoat.

The trees grow to 20 m and have a dark scaly bark. Arising from underground horizontal roots will be structures called pneumatophores which resemble long pencil like straws bunched under the canopy and often extending some distance from the parent tree. These pneumatophores serve as aerating branches or roots for gas exchange in the anaerobic mud the Black Mangroves often live in. There is evidence that indicates the plane of the mean height of the Black Mangrove pneumatophores may well approximate a biological indicator of the mean high water mark (MHW). How might you prove or disprove this idea? The Black Mangrove detritus is generally "pulsed" into the estuarine food chain only during the highest flood tides of the year and during storm periods when Black Mangrove peat is released into the bays and seas with its special set of nutrient components.

THE WHITE MANGROVE—*Laguncularia racemosa*

The White Mangrove and Buttonwood are the least salt tolerant of the mangroves and are usually found growing in higher elevations where the soil or substrate is less saline.

The White Mangrove grows to 20 m or more and has fleshy, waxy, ovoid, light green leaves with two salt pores, or glands on the leaf petiole (leaf stem). The leaves are arranged on the stem in an opposite fashion. The flowers are small white and bell shaped. The seed is about 1.0-1.5 cm slightly fleshy, and broadest at the apex. The seed may enlarge and begin to germinate while on the parent or floating in the water.

The White Mangrove delivers its detrital based nutrients in a variety of pulsing patterns depending on location. If along streams and rivers it may deliver daily but if upland of Black Mangrove in the high marsh it may deliver similarly to the Black Mangrove pattern.

BUTTONWOOD—*Conocarpus erecta*

The Buttonwood is usually found growing in the same area as the White Mangrove. The Buttonwood belongs to the same family (Combretaceae) as the White Mangrove.

The tree will grow from 15-20 m tall with smooth small (2-7 cm) leaves arranged in an alternate fashion on the stem. Like the White Mangrove, the Buttonwood has two small salt glands on the leaf petiole at the base of the leaf. The flower forms a conelike green head 1-2 cm wide. The fruit becomes a leathery brown button-like cone that gives the tree its name. Detritus circulated similar to White Mangrove.

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RED MANGROVE
(Rhizophora mangle)

BLACK MANGROVE
(Avicennia germinans)

WHITE MANGROVE
(Laquncularia racemosa)

BUTTONWOOD MANGROVE
(Conocarpus erecta)

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TIDES AND THE 24 HOUR CLOCK

PART I. THE 24-HOUR CLOCK

INTRODUCTION

Tides are important to anyone living near the sea. The effects of the tides and the causes of the tides are fairly well-known. Fisherman, pilots, marine researchers and even a field trip for a marine science class depend on the knowledge of local tides. To properly read tide tables and avoid confusion, the 24-hour clock is used. We shall consider the 24-hour clock first, to be followed by excercises involving the tide tables.

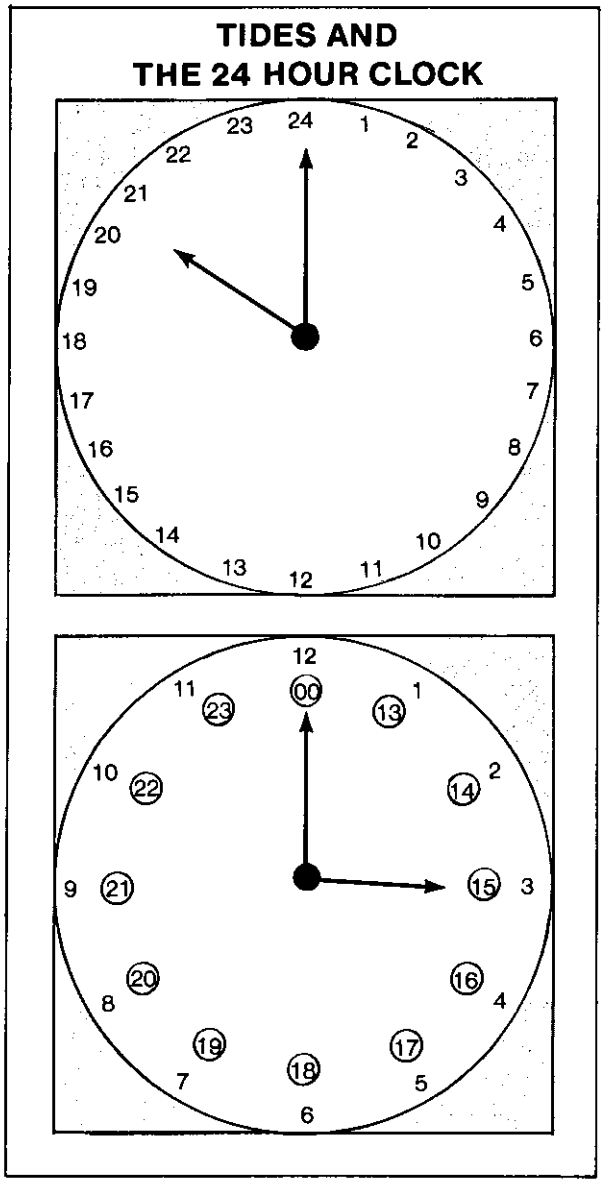


FIG. 1

TO THE TEACHER

Use a 24-hour clock if it is available. If not then make a model or place "stick-on" labels on a standard 12-hour clock. Mark the hours 13 to 00 as shown by the diagram (Fig. 1). To give the students a grasp of the 24-hour clock, take a globe and view it from one of the poles, illustrating that the earth revolves once in 24 hours and is a large clock.

TO THE STUDENT

There are many reasons for using a 24-hour clock. In using the 24-hour clock there is no need for the terms AM or PM. There is only one possible time for a given designation on the 24-hour clock. The 24-hour clock is adaptable to Greenwich Mean Time (GMT) or Universal Time (UT) for navigation and worldwide usage. Modern communications demand the use of UT. The U.S. Dept. of Commerce Tide Tables uses the 24-hour time exclusively as do many navigational charts and tables.

PURPOSE

To learn to use the 24-hour clock.

MATERIALS

24-hour clock or model
Paper and Pencil

EXERCISE I

Conversion to 24-hour time

12-hour clock	24-hour clock
1. 7:52 AM	_____
2. 7:52 PM	_____
3. 10:00 AM	_____
4. 10:00 PM	_____
5. 3:30 PM	_____
6. 5:15PM	_____
7. 12:00 noon	_____
8. 12:00 midnight	_____
9. 12:04 AM	_____

Conversion from 24-hour time

24-hour clock	12-hour clock
1. 1450	_____
2. 0524	_____
3. 1641	_____
4. 0032	_____
5. 2410	_____
6. 1947	_____
7. 2001	_____
8. 0629	_____
9. 1006	_____

EXERCISE II

Computing time differences

Note: The first two digits refer to hours and last two digits refer to minutes. When adding if the last two digits are more than sixty you must convert these minutes into the appropriate hours. When subtracting you must borrow an hour, be sure you convert that hour into 60 minutes. NOT 100. How would you indicate seconds in 24-hour time notation?

	Addition	Subtraction
a)	0240 hrs +0120 hrs	1358 hrs −0050 hrs
b)	1708 hrs +1000 hrs	0911 hrs −0119 hrs
c)	2250 hrs +0120 hrs	0234 hrs −0050 hrs
d)	1100 hrs +0230 hrs	0120 hrs −0300 hrs

PART II. TIDES

INTRODUCTION

Tides are waves of water with an extremely long period caused by the gravitational effect of the Moon, Sun and Planets. Due to the rotation of the earth, tides generally occur twice a day (semi-diurnal). In some locations the tide occurs only once a day (diurnal) or an irregular mixture of both. The mass of water moved by the tides is truly phenomenal. The Bay of Fundy tides alone move more than 100 billion tons of water in and out with each tide. In comparison the Mississippi River needs 140 days to move the same volume of water. Accurate predictions of tide times and heights are important to all who work, live, or play near the shore.

The average time between the complete tide cycle is 12 hours and 25 minutes. When the Sun and the Moon are aligned, new moon and full moon, the tidal variation is at its greatest; this is called *spring tide*, (no relation to seasons). During the first and last quarter of the moon the gravitational pull of the Sun reduces tidal effect of the moon. However that gravitational effect of the moon is the predominant force responsible for tides. Tides during the first and last quarter are called *neap tides*, the variation between high and low tides is less than at any other time.

The seiche (oscillating or sloshing of enclosed bodies of water) periodic winds, variations in atmospheric pressure, rainfall, ocean topography and currents all are superimposed on the measured water level at coastal locations, which are predominantly tidal fluctuations. The most to be expected from a tide table is that it will be correct in calm weather with a steady barometer (Darwin, 1962).

Under certain conditions the tide will cause a wall of water to rush upriver in a spectacular show. The Petitcodiac River at the head of the Bay of Fundy is just one of several coastal locations where this phenomenon occurs. The upriver rush is called a *tidal bore*. On the Amazon a tidal bore is called the *Pororoca* and looks like a long waterfall, up to 16 feet in height, moving upriver at speeds up to 12 knots. The roar from this bore is often heard as far away as 15 miles.

A so-called tidal wave is not caused by the gravitational pull of any celestial body. It is *not* a "tide" condition at all. The Japanese term *Tsunami* (soo-nah'-me) is used by marine scientists in referring to these sudden increases in water level, which are caused by earthquakes or submarine disturbances.

EXERCISE I

The *Tide Tables* contain seven different tables. For this exercise we will only be concerned with Table 1 — *Daily Tide Predictions* and Table 2 — *Tidal Differences and Other Constants*. Before you use these two tables you should read the "Explanation of Table" at the beginning of each Table. This clearly explains the meaning of the data and will make using the table much easier.

A. Find the times of high and low tides for Miami Harbor Entrance, Florida for October 26th, 1980 (or whatever year *Tide Tables* you have).

Step 1. — The *Tables* are arranged starting with the northern most places of prediction down the Atlantic Coast around the Florida Keys then up the West Coast of Florida covering the Gulf states. Find Miami Harbor Entrance, Florida.

Step 2 — The months are arranged three to a page. Turn to the page showing October, November, and December. Now, going down the October column, find October 26th and record the four lines of data (Times and Heights). Determine the times of both the high and low tides by noticing the change in height.

B. Find the times of high and low tides for Ponce de Leon Inlet, Florida for October 26th, 1980.

Step 1. — Since Ponce de Leon Inlet is not listed in Table 1, we must go to Table 2. Near the end of the book you will find the "Index to Stations" which are listed alphabetically. Find Ponce de Leon Inlet, Florida and record the number.

Step 2. — Turn back to Table 2 and find Ponce de Leon Inlet. Read across to "Differences, Time." The High water time difference reads: plus (+) 0 hours and 06 minutes. The low water time difference reads: plus (+) 0 hours and 20 minutes. This difference is applied to Miami Harbor Entrance which is the reference station.

Step 3. — Refer to Miami Harbor Entrance for October

26, and apply the correction for Ponce de Leon Inlet. You should now be able to report the correct predicted times for high and low tide at Ponce de Leon Inlet, Florida.

You may also note there are "Differences in Height" which should be applied to the reference station. Try doing two or three other stations and dates. This will give you practice in using the Tide Tables.

There are a couple of variables that the tide tables don't consider. Normally Daylight Saving Time is not allowed for, so in the summer you need to add an hour. The other variable is even trickier, for it is a continuum of times, applicable to points along the rivers back from the inlets. To find how much lag there is from the ocean to where you are, you can ask commercial fishermen or boaters or can check tide times for yourself. Do this several times, to obtain a good average.

EXERCISE II

Construct a tide chart for several days using data from a prediction table.

Plot from prediction table below.

TIDE PREDICTIONS

DAY	TIME		HEIGHT
	HOURS	MINS.	FEET
1	01	15	1.0
	07	30	4.1
	16	00	1.0
	20	15	4.0
2	02	00	1.0
	08	30	6.5
	14	15	2.0
	21	00	5.0
3	04	15	0.0
	16	00	7.0
4	04	30	1.0
	10	00	3.0
	16	00	1.0
	22	00	2.9

TIME 24 HOURS (00-24) MIDNIGHT TO MIDNIGHT — HEIGHT MEASURED FROM MLW (0 REFERENCE LINE)

First day has been plotted, complete days 2, 3, 4 and try to answer the questions from the chart.

1. During what day is the tide diurnal? _____
2. During what day is the tide mixed? _____
3. During what day is the tide semi-diurnal? _____
4. During what day is the tidal range largest? _____
5. What is the smallest range and on what day? _____ft. _____day.
6. What is the largest diurnal inequality shown? _____ On what days? _____ and _____.

EXERCISE III

Charting Tidal Characteristics

Purpose: This exercise is intended to help the student see the differences in the types of tides and their ranges along the southeastern U.S. and Gulf coasts.

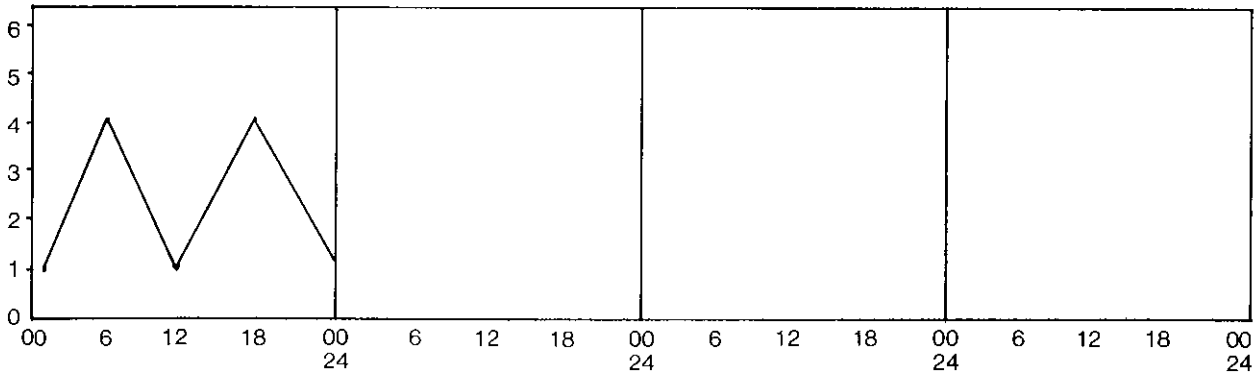
Materials: Graph paper, colored pencils (pens), ruler or French Curve.

Procedure: Using standard graph paper follow procedure described in the previous exercise. Plot two full tidal cycles for 6 locations on the Atlantic coast and 6 locations on the Gulf coast. Label each graph (location) and determine if the tide is diurnal, mixed, or semi-diurnal. Label on graph, then show on accompanying chart, using a color code, the extent of each type of tide along the Gulf and Atlantic coasts from Brownsville, Texas to Norfolk, Virginia.

QUESTIONS FOR CONSIDERATION:

1. What location on the Atlantic coast has the greatest tidal range? Which has the lowest?
2. What different types of tides are observed on the S.E. Atlantic coast? Why does this occur?
3. In the Gulf of Mexico what are the maximum and minimum ranges?
4. Along the Gulf coast what different types of tides are observed? Why does this occur?
5. Are there advantages to a large tidal range for a port city? Explain.
6. For fishing or recreational beach development which would you prefer, a large or small tidal range? Would the type of tide effect your preference? Why or why not?

TIDE CHART



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Tides and Current—1958, 16mm, color-sound, 15 minutes.

Ocean Tides: Bay of Fundy—1956, 16mm, color-sound, 15 minutes.

CHARTING LOCAL CURRENT SYSTEMS

INTRODUCTION

Currents along beaches, inside local inlets and passes and inside bays, lagoons and estuaries have become increasingly important as human development has concentrated on waterfront property because of its high profit potential to developers and landowners. Generally coastal developments have taken place with little consideration or knowledge of the normal changes that occur on dynamic beaches and with little recognition of the resulting biological effects caused by drastic changes of circulation in shallow inshore waters. The ultimate effects of such developments might include loss of buildings to beach erosion, loss of biological communities, increasing eutrophication and siltation of formerly clean and clear waters, the buildups of undesirable sediments often requiring the costly removal at large public costs. A knowledge of local current systems, their fluctuations and their influence in changing the "land-water edge" might reduce or eliminate poorly designed coastal developments. This knowledge can also provide useful information for consideration by planners, developers, conservation groups and prospective buyers. A well done class project with accurate data and a comprehensive historical background can be useful for future reference in the making of important decisions.

"Charting local current systems" means just that. We do not intend in this unit to chart the North Atlantic Gyre or the Gulf Loop Current, but will confine our examination to those currents which influence a few miles of local shoreline. These local studies are important to many groups and agencies, including, but certainly not limited to: developers, the Florida Department of Natural Resources, Environmental Regulation, Planning and Transportation, The U.S. Army Corps of Engineers, local planning agencies, conservation groups and others. Commercial and sport fishermen, boaters, swimmers, surfers, chartmakers and beach erosion experts are a few of the private individuals whose lives or work are affected by local current systems. Local changes in these systems are a major concern for every coastal resident.

Now for a few definitions: *Tide* is the rise and fall of a large body of water, such as an ocean, due to changes in the gravitational pull of the moon and the sun on the water mass. A *tidal current* is the horizontal movement of the water. As the tide rises and falls, the currents of water move toward the shore and away from shore so we say the tidal current floods and ebbs. A *nontidal current* is any current not due to the tidal movement. *Near-shore currents* are the sum of tidal and nontidal effects and are most complicated. They are the result of many different agents acting on the water: the direction, force and duration of the wind; the rise and fall of tides; the constantly changing shape of the bottom of the near-shore water; upland runoff and the sediment brought down by rivers and streams which in turn affects bottom currents. *Estuaries* are bodies of water where upland runoff and seawater mix. They are mostly surrounded by land, with a direct tidal connection to the sea. Their complex current systems are very difficult to study.

Many methods are in use to determine local current systems. The oldest and still the most widely-used method is the use of drift bottles. This method has been extremely valuable in measuring offshore ocean currents, but is not too satisfactory near shore. In charting nearshore and estuarine currents, dyes of various colors are dropped into the water and visual observations are made. Another indirect way of measuring bottom currents is to dye sand and determine its drift. With the increase in surfing all over the world, personal observations of local currents have been made. As waves and currents cause beach erosion, specialists in this field (Corps of Engineers, Scripps Inst. in California and others) have made valuable contribution to our knowledge of currents. Changes in the shape of the beach often give very accurate indications of current velocity and direction.

TO THE TEACHER

In order to simplify, local current systems are divided into three categories: Beach Currents, Jet Currents through inlets and passes, and Inshore Currents in estuaries, bays and lagoons.

TO THE STUDENT

The measurement of currents is easy to do, but sometimes hard to believe. Most people overestimate the speed of moving water, often doubling or tripling its real speed. You will have to watch out especially for wind effects on your floats, and you will have to use enough floats to get a good average.

Generally, current studies should be done through several complete tidal cycles to achieve good average figures. Winds effect currents too, and studies should continue through several seasons to account for prevailing wind changes.

FIELD PROCEDURES FOR MEASUREMENT OF NEAR-SHORE CURRENTS

1. Use of floats outside the breaker zone, floats can be made by students.

Paint plastic milk jugs with marine resistant phosphores-

cent orange. Fill with enough sand to float just awash. Seal the top. A stiff wire such as a coat hanger can hold a small numbered or colored flag. The sketches give some suggestions on the construction of vane floats and milk jug floats. The effect of the wind should be reduced by having as little of the float above the water as possible. Currents beyond the breaker zone are often weak, so there should be a large float surface for water reaction. Floats should be released along the entire length of the area to be studied. Each float should have an identifying flag or marker.

2. Ordinary rubber balloons filled with fresh water may also be used, balloons are best filled at a faucet. A permanent felt marker can be used to mark balloons. They are put into the water beyond the surf zone where they float, due to the fresh water having less density than the salt water. Grapefruit have also been effectively utilized in this same manner.

3. The use of flourescein dye for the measurement of currents inside the breaker zone - Flourescein is a yellowish-red dye which receives its name from the brilliant yellowish-green fluorescence of its alkaline solutions. A cup of sand with a teaspoonful of dye is wrapped in a paper towel, bound with a rubber band and tossed into the breaker zone. The direction of movement of a patch of colored water is then traced from shore, from a pier if one is nearby, or from a boat.

BEACH CURRENTS

Generally, there are two types:

Longshore - active in transporting sand in the breaker zone, caused by the waves hitting the beach at an acute angle.

Vertical to the shore - the return flow from waves and tides cause rip or runout currents which may be dangerous to bathers.

These currents are easy to measure. You may use floatable material picked up on the beach for a "quick and dirty" approximation of direction and speed, and then use prepared floats for a comprehensive study. Currents within the breaker zone are usually zig-zag, in and out with the waves and a movement along the shore in the direction sand is being transported.

JET CURRENTS

Narrow inlets and passes constrict the downhill flow of the tidal waters into a high velocity "jet" which then enters relatively still water and spreads out. The amount of spread (plume) for a typical tide change corresponds to a semi-circle with a radius approximately 500 times the mean depth of the inlet or pass. The speed of this current may exceed 5 knots in many local passes and inlets. In Storstraum Channel, Norway speeds of 16 knots (18.5 mph) have been recorded during spring tides.

Floats thrown from shore are generally sufficient to measure these currents. Make measurements at several stages of flood and ebb, as current speed varies hourly.

INSHORE CURRENTS

In many coastal areas and estuaries rotary currents are observed. These are caused by the rise and fall of the tide and in many places completely mask other local or wind-induced currents. In wide estuaries, where flow is not restricted, the current direction changes through all points of the compass. A boat anchored under these circumstances will point in every direction at some time during the day.

Investigate these current systems using floats not affected by the wind. Aerial photos of the area may aid in your investigation. Pay special attention to the position of sandbars in shallow water which may impede or alter water flow.

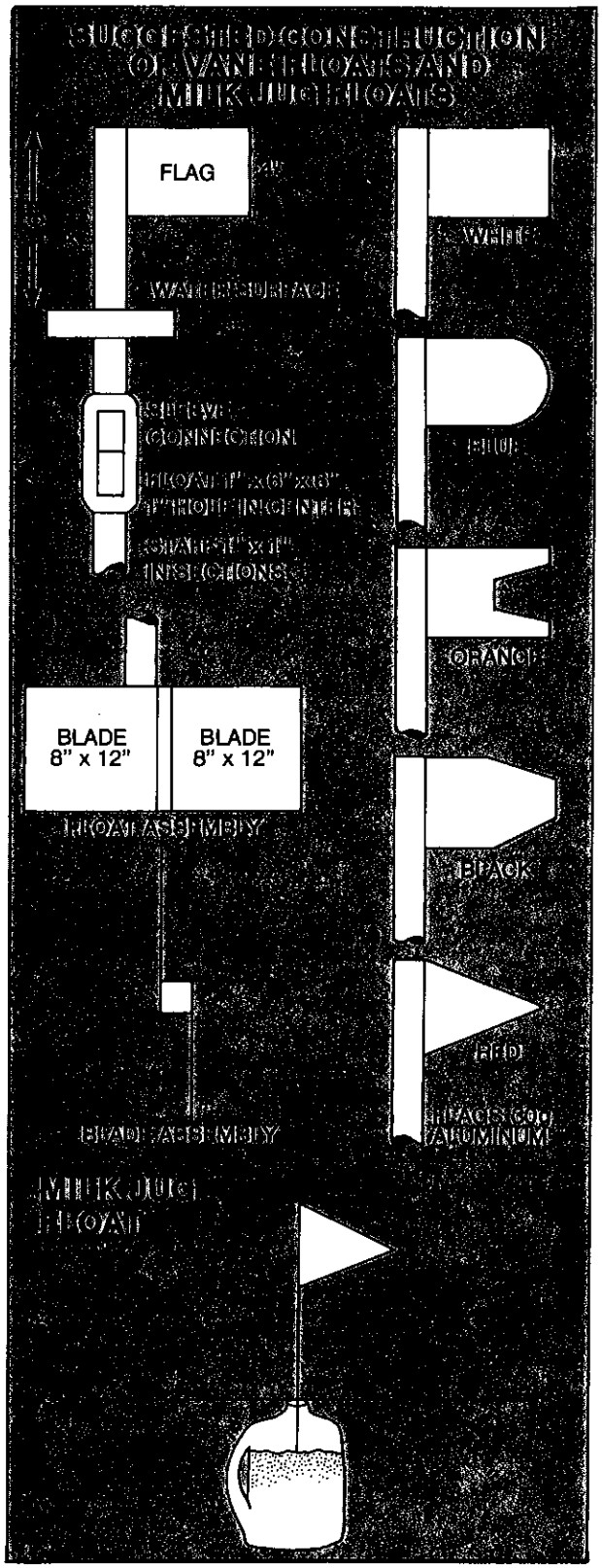
QUESTIONS FOR CONSIDERATION

1. Describe and diagram a typical "run-out" or rip current on a beach. Describe what a swimmer should do to escape one.
2. Why might surface currents near shore be different from bottom currents?
3. What effects would a jetty or groin have on local longshore currents?
4. Diagram typical current flows for nearby inlets, passes, beaches, river mouths. Indicate current speed and direction, time related to tide. A current rose such as on a nautical chart may be useful.
5. What are the differences between tidal and nontidal currents?
6. Why is the mouth of an inlet or pass generally very choppy on the ebb but not on the flood?
7. Design a waterfront residential community with a "full-flushing" canal system. Hint: think in terms of tides, inlets and basins. Make a model to prove your idea.

CONCLUSIONS

It is easy to see that the measurement of near-shore currents is very difficult. Much work of a quantitative, statistical nature needs to be done. Every marine science education group should have a master chart of its own coastal area and as current studies are made, this should be put on the chart. The knowledge of currents which surfers, swimmers and fishermen gain from experience should be recorded for the benefit of everyone. For a group wishing to make a really serious study of the beaches, monthly charts should be prepared showing changes in the beach topography due to current, wave, and wind action. The study of sand transport along the beach is most interesting and could be a real contribution to the community.

Photos of the same area from the same vantage point over a period of time may be used to show the effects of currents on shorelines, bars and channels.



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Films: "The Beach, A River of Sand" E.B.F., 15 min., color. "Waves on Water" E.B.F., 15 min., color.

BEACH ANALYSIS

TO THE TEACHER

The newspapers are constantly featuring articles whose themes involve the disappearance of beaches. In Florida, the Miami Herald, Palm Beach Post-Times, Tampa Tribune, and Sarasota Herald-Tribune are noteworthy in this respect.

It is suggested that students might bring in current articles before the exercise is scheduled. The political and economic significance of beaches and the recreational use values should produce lively discussions.

This exercise can and should be a part of the general field trip. While some students collect biological specimens, others can collect sand, examine the sedimentation pattern and map the collection sites. If there is an extensive section in the program concerning the geologic aspects of beaches and sub-surface cores, an entire geologic field trip can be justified.

Beach profiles made several times through a year can easily document seasonal changes. Photographs help, but don't replace the usefulness of profiles. "Nests" of sieves are useful in sand grain analysis and can be obtained from any scientific supply house or a simple design and a friendly shop teacher could work wonders. Part VI is best done while the overview is in progress.

TO THE STUDENT

The facts that the beaches are disappearing, unavailable and crowded, are unquestioned. Knowledge of the size and types of sand particles and the factors affecting the deposition and removal of particles is important. Knowledge is best founded on experimentation and factual data.

This exercise has several parts. A successful exercise depends upon each getting proper consideration.

MATERIALS

Compound Microscope (dissecting)
Magnifying glass
Coffee can with both ends removed (1 lb. size)
Range pole
Sieve set
Compass
Small shovel
Shallow pan for drying
Samples
Hand level (Abney)
Camera (Instamatic)
Hand scoops (assorted)
Square of plywood to hold map flat
Plastic bags
Petri dishes
Probes
Small paint brush with moderately stiff bristles (½" - 1" wide)
Rule
Scales

STATEMENT OF THE PROBLEM

To recognize, record and analyze beach characteristics, the problem is divided into six parts. They are:

Part I - A. The overview
B. The beach profile
Part II - The cross-section profile
Part III - Sampling procedure
Part IV - Moisture determination
Part V - Particle sizing and identification
Part VI - Wave Analysis

Part I

A. The overview: While standing on a promontory from which a large section of beach is visible: (1) select sampling sites; (2) map the intertidal zone; (3) map the line of vegetation; (4) circle the areas of similar vegetation. Next, plot obvious deposition characteristics such as: (1) an area of shells; (2) driftwood; or (3) drift algae. For mapping techniques refer to the attached beach characteristics sketches.

B. The beach profile: The profile-taking party should consist of three persons: a rodman, an instrument man and a recorder. Typically their duties are defined below:

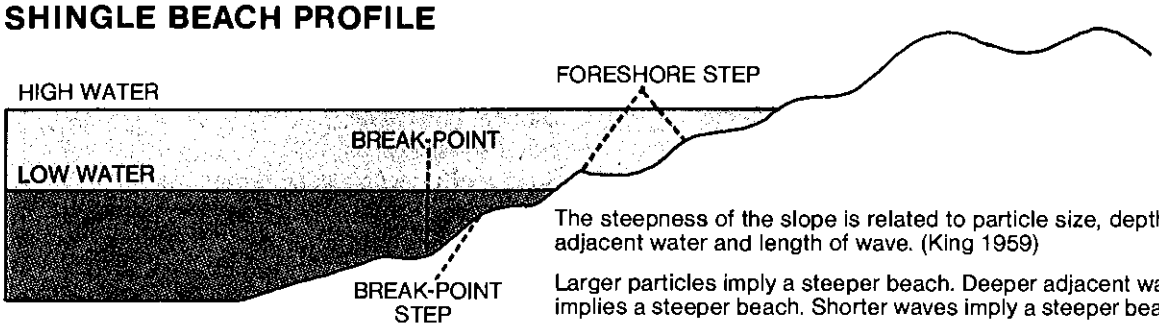
The *rodman* holds the rangepole on designated interval points as he traverses a line perpendicular to the beach so that the instrument man can sight the elevation of the points (heights above mean low tide are positive). Rodman can either pace a regular distance between points (say 10 meters) or he can pace the metric distance between prominent "breaks" in elevation. Do NOT change systems in mid-transit, however.

The *instrument man* reads the elevation while holding the instrument level. He then calls the elevation above mean low tide (+) or below mean low tide (−).

The *recorder* relates the map plot to the location of the range pole, records the elevation and then locates the next map plot in anticipation of the next reading.

MAJOR BEACH CHARACTERISTICS TO RECOGNIZE

SHINGLE BEACH PROFILE

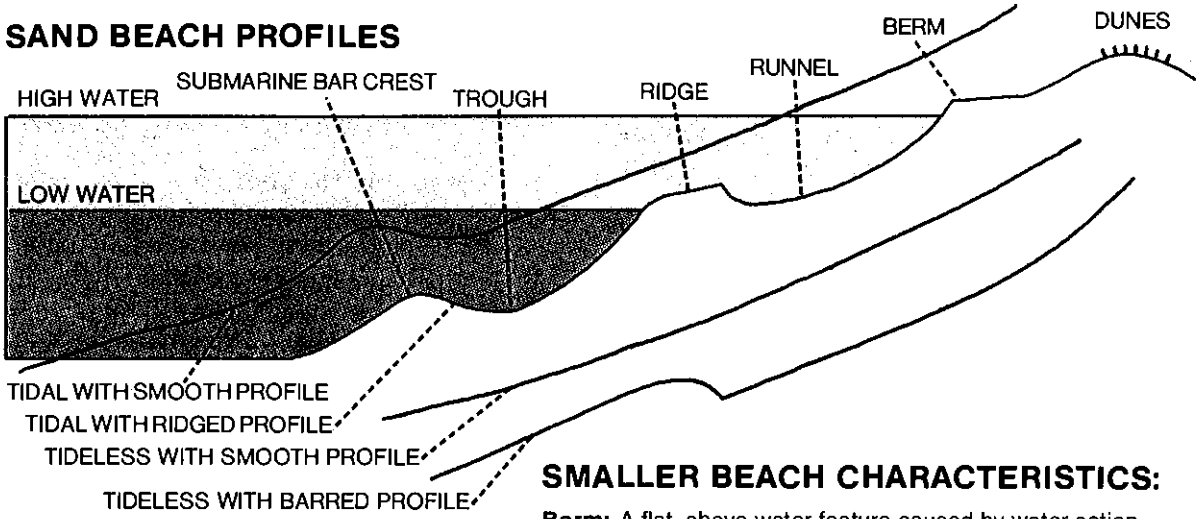


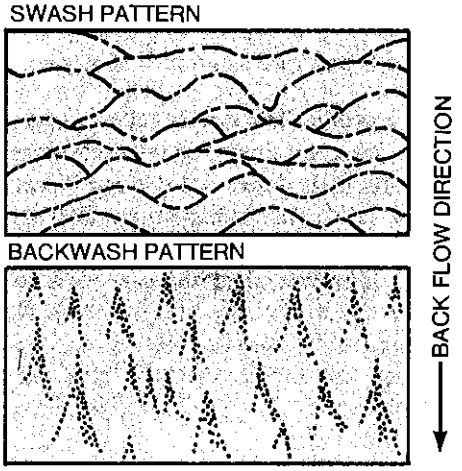
The steepness of the slope is related to particle size, depth of adjacent water and length of wave. (King 1959)

Larger particles imply a steeper beach. Deeper adjacent water implies a steeper beach. Shorter waves imply a steeper beach.

Shingle Beach: the slope is steep, similar in slope to the slope of a pile of sand poured from above. A step is usually found at the low tide line.

SAND BEACH PROFILES





SMALLER BEACH CHARACTERISTICS:

Berm: A flat, above-water feature caused by water action.

Bar: An underwater ridge of sand usually parallel to the shoreline.

Wave Frequency: The number of waves per minute.

Wave Velocity: Speed of wave.

Wave Length: Distance between crests.

Cusp: A series of evenly spaced crescent shaped depressions below the berm.

Swash: The last rushing water from a wave is a thin sheet. The successive swash marks appear as overlapping crescents.

Backwash Pattern: When retreating water passes over minute projections downward, delta (△) shaped marks are left.

Sand domes: As water percolates through sand, entrapped air escapes, causing dome shaped bulges. The domes usually have pinholes in them.

Ripple Marks: Parallel ridges and troughs.

Part II - The cross-section profile

A. To profile a cut, bluff, dune or other nearly vertical surfaces:

1. Clean debris from the face of the surface. Use a sweeping motion of the shovel's edge to cut a smooth, vertical plane. All strata should show in profile.

2. Place a measuring device (range pole, meter stick, or a common object of known size such as the shovel) in a true vertical line against the cleared face.

3. Photograph the strata and measure.

4. Select interesting strata for sampling (See Part III-B).

B. To profile a flat.

1. Excavate a hole with a width greater than the closest focusing distance of the camera, otherwise the picture will be out of focus. If the hole shape is deltoid (▽) less sand needs to be excavated.

2. Follow steps 2, 3, and 4 above.

3. Refill the hole!

Part III - Sampling procedure

A. Surface Sampling: Sampling should be done at several sites along the profile or transect. Before taking the sample, remove any surface debris.

1. Push the opened coffee can "corer" flush with the ground surface.

2. Remove the earth, sand, pebbles, shells, etc. with scoops. The contents should go directly into the plastic bag. The excavation should be carefully made to the exact level of the "corer" bottom.

B. Strata sampling: (1) Once a stratum is located (See Part II), push the "corer" into the stratum until it is flush with the surface. Care must be taken to insure that the can's contents are entirely from the selected stratum. (2) Excavate, bag and tag the samples as before.

BACK IN THE LABORATORY

Part IV - Moisture determination: The moisture-holding property of earth is an indicator of the amount and types of life it can support.

A. Determine the mass of the sample.

B. Determine the mass of a shallow pan.

C. Spread the contents of sample onto the pan.

D. Dry the pan of sample in a household oven at 150° - 200°C (300° - 390°F).

E. Determine the mass of pan and contents after several hours.

F. Replace in oven for one hour. Determine mass again. If the last determination is the same (D), the sample is dry. Otherwise return to oven until successive readings are the same.

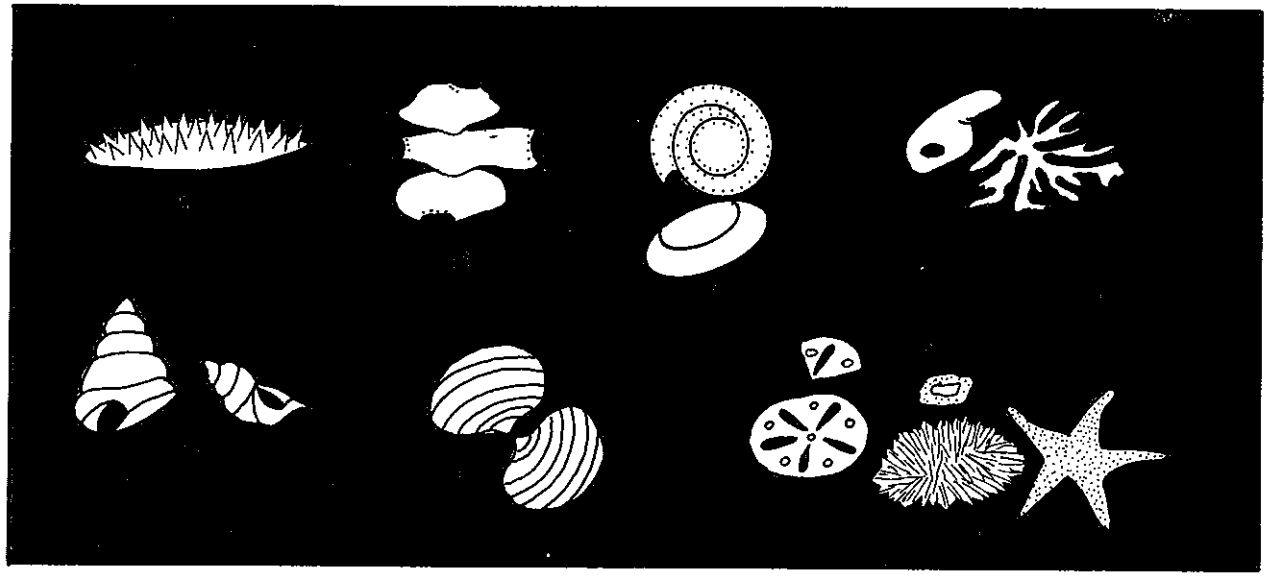
G. Mass of water: "A" + "B" − "FF" = mass of H_2O (the letters refer to data from these lettered steps).

H. Percent of moisture:

$$\frac{\text{"G"}}{\text{"F"} - \text{"B"}} \times 100 = \%H_2O$$

Part V - Particle sizing and identification: Arrange the individual sieves in a nest such that those with the largest sieve opening are at the top. Each sieve in the set has a smaller opening than the one above. Finally there is a bottom pan and a cover. A typical sieve set comprises 5 or 6 sieves. NBS

GRAIN GUIDE



A. Alcyonarian—The sea fans and sea whips. The skeletal spicules of this group are often linked to elongated footballs with short spines. They appear in various shades of reds and purples, and are very frequently opaque.

B. Halimeda—A green algae with a calcareous skelton. The plates are flat and often show the holes for attachment. They may show signs of chlorphyll if they are fresh.

C. Foraminifera—A group of calcareous protozoa. A wide variety of shapes, oval, flat plates frequently with exposed pores.

D. Corals—Often very large coarse fragments which show the individual polyps septa and the colonial structure.

E. Gastropoda—The group of mollusks which possess a single, spiralling shell, as in conchs, tulip shells.

F. Pelecypoda—Mollusks with two shell parts as in clams and oysters.

G. Echinodermata—The urchins, sea stars and sand dollars. These produce a variety of grains including the urchins spines, fragments of the exoskelton and flat pieces of dollars.

Date:_____ Sample No._____

Location:_____ Moisture Content (%)_____

Experimenter:_____ Amount of dry sample_____

**Granule as a size class may be omitted. Either "gravel" is substituted or the 2-4mm size range is included in "pebble"

***The particle shape fraction percentage may also be found by spreading a representative amount in a shallow dish, then count and classify 100 particles. The number in each shape classification is then the percent (see statistics)

Rounded Subrounded Angular Disclike Faceted

CHART #1
NON-CARBONATE SANDS

A	B	C	D	E	Particle Shape Fraction
Sieve Size	Particle Size Classification	Size Limits	Mass	$\left(\frac{\%\text{mass of fraction}}{\text{mass of sample}}\right)$	$\left(\frac{\text{Mass of shape fraction X 100***}}{\text{Mass in Column D}}\right)$ percent
Measurement by Rule	Boulder	Larger than 256mm (10")			Fossils Animal cast (shells)
Measurement by Rule	Cobbles	64mm (2.5")			
7	Pebble	4mm			
10	Granule**	2mm			
230-250	Sand	1/16mm			
See Measuring with a Microscope	Silt	1/256mm			
Same as above	Clay	Smaller than			

Date:_____ Sample No._____ Experimenter:_____

Location:_____ Moisture Content (%)_____ Amount of dry sample_____

CHART #2
CARBONATE SANDS

A	B	C	D								
Sieve Size	Size Limits	Mass	$\left(\frac{\%\text{mass of fraction}}{\text{mass of sample}}\right)$	ALGAE	FORAMINIFERA	CORAL	ALCYONARIAN	GASTROPODA	PETCYCEPODA	ECHINODERMATA	MISC. CO_3
Measurement by Rule	Larger than 256mm (10")										
Measurement by Rule	64mm (2.5")										
7	4mm										
10	2mm										
230-250	1/16mm										
See Measuring with a Microscope	1/256mm										
Same as above	Smaller than										

(National Bureau of Standards) sizes used for selection are 5,7, 10, 35, 80, 250 and 325.

A. 1. Scoop dried sample onto the top sieve. 2. When the pile of material indicates that the sieve is blocked, put on the top; rock the nested sieves to-and-fro. Allow the bottom to "bump" on each movement. Allow the whole stack of sieves to turn in the hand after each oscillation. 3. Periodically add more material to the top sieve until all of the sample has been graded for size. If one or more sieves or the bottom pan fills with material; this can be emptied into a bag(s) or container(s). The stack of sieves can be reassembled for continued operations.

B. Sample removal and appraisal: 1. Remove cover. 2. Agitate the top sieve to cause the sub-sized material retained on this sieve to pass through the openings. Stirring with the handle of the brush will h elp to cause these particles to pass through. 3. Empty the contents retained on the top sieve into a separate container of known mass. 4. Determine mass of container plus sample fraction. Subtract mass of the container. 5. The percent (%) is found by dividing the mass of fraction by mass of whole dry sample. Repeat steps 1 - 5 above for each succeeding sieve.

C. Use a sample of sand weighing one gram from each sieve size, each grain is to be identified and counted. Place the gram sample in a Petri-dish and using the compound microscope begin by simply identifying the grains. For Non-Carbonate sands record your data on Chart #1 noting various particle shapes. For Carbonate sands record your data on Chart #2. The Grain Guide will assist you in identifying the origin of carbonate particles.

Part VI - Wave Analysis

A. Determine the frequency of the waves in number of waves per minute. Once this is known, stand at the water's edge and count out the same number of crests. Ex. F= 4/minute. Then count four crests outward.

B. Tilt hat forward or place hand above the eyes. Palm down as if to shade eyes until the 4th (example) wave is at bottom edge of hat or hand.

C. Rotate head to left or right - sight a spot on the beach on bottom edge of hat or hand.

D. Pace distance to that spot. (This is an old Boy Scout trick to measure the width of a stream.)

E. This is roughly the distance traversed by a wave in one minute. The velocity of the wave is shown in $\frac{\text{meters}}{\text{minute}}$

F. Since we also know how many waves are inside the distance, a division by frequency equals wave length.

G. Wave heights can be estimated by lining up the crests with the horizon using your eye to sight with and measuring to the water level at your feet.

QUESTIONS FOR CONSIDERATION

1. Does the general appearance of a beach change with the seasons? If so, why? If not, why not?
2. Could the gain or loss of beach material be computed by comparing beach profiles from time to time?
3. Could the roundness of particles be related to the "age" of the particles?
4. Sand particles from a single stratum are:
   a. The same size and shape?
   b. Vary in size but same shape?
   c. The same size, but shape varies?
   d. Vary in size and shape?
5. In carbonate sands, what groups of organisms has produced the greatest percentage of carbonate grains in the sample? Is this the most common group from the area? Where were the grains produced? How were they transported?

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INTRODUCTION TO THE ANALYSIS OF SEA WATER

TO THE TEACHER

We teachers sometimes make the mistake of teaching labs in a vacuum without relating the objective of the lab to living kingdoms.

Because of the increasing stress man has placed on the sea as a habitat, it is now more important than ever that marine studies assist students to understand the ecological role that both physical and biological factors play in support of marine ecosystems. It, therefore, becomes increasingly important to demonstrate how each lab activity relates to the sea's life support system.

Of what value is it to measure seawater salinity, if we don't also relate the ecological effects of the determined salinity to the flora and fauna? Also, what effect does human activity have on altering the salinity? Once these questions are answered, we must then look at possible short and long term problems created and the solutions and/or modifications necessary to correct any negative changes.

TO THE STUDENT

As a young child, did you ever go to the beach with your family for a day of fun in the sun? Did you ever capture a little sea creature such as a fish, crab or snail? You probably placed it in some seawater in a plastic pail or paper cup and proudly announced to your parents that you were going to take it home as a pet. More than likely by the end of the day your little creature died. Why did it die? It was in seawater and sea animals live in seawater.

All too often, man looks at life in the sea in this isolated fashion, believing that as long as there is food to eat and water to live in the creatures in the sea can live.

In order for us to promote a healthy marine environment, we must understand the ecology of the sea far beyond the "plastic pail" philosophy. We must ask ourselves why animals are found where they are. What are their environmental demands? What is their tolerance to the surroundings?

The limits of tolerance describe a range of living conditions specific to each organism and each living or non-living factor effecting it. The range of tolerance of a limiting factor may be very large, or very small. Killifish tolerance to salinity is great, but the Everglades Kite is limited to just one food organism, the apple snail.

Every species of plant and animal has a range of tolerance for each factor which determines its survival. There are tolerance limits for food, predation, shelter, breeding conditions, temperature, salinity, water clarity, light, pressure, oxygen, depth and other factors that effect its survival at any stage of a life cycle. An animal's survival is often limited by the factor with the narrowest tolerance. This can be described as the Law of Minimums.

OBSERVATIONS:

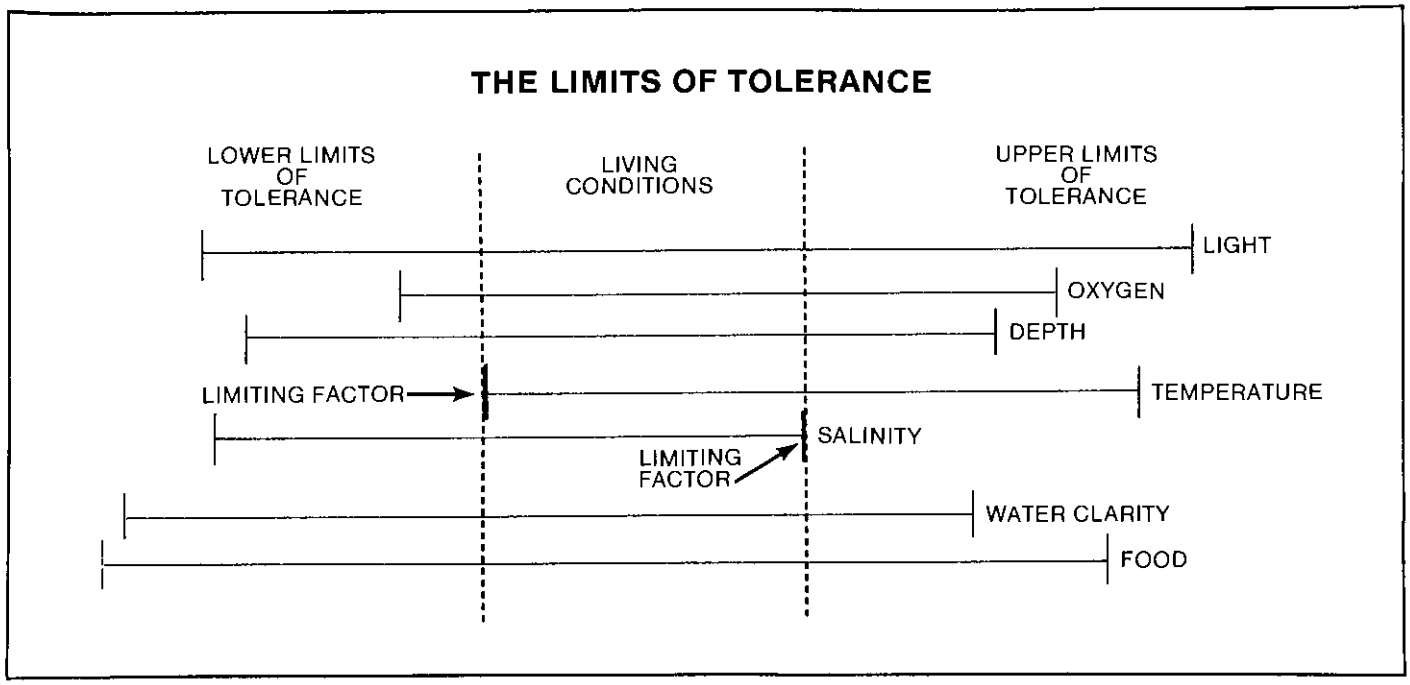
1. Organisms may have a wide range of tolerance for one factor and a narrow range for another.
2. Organisms with wide ranges of tolerance for all factors are likely to be most widely distributed.
3. When conditions are not optimum for a species with respect to one ecological factor, the limits of tolerance may be reduced with respect to other ecological factors.
4. The period of reproduction is usually a critical period when environmental factors are most likely to be limiting.
5. There is a decrease in the number of organisms near the limits of tolerance for each factor.
6. Organisms may have differenct limits of tolerance in different parts of their life cycle.

Factors we study in marine science either directly or indirectly relate to a marine organism's ability to tolerate and survive and its environment. Do you understand now why the little marine pet did not survive in the plastic pail? Is it possible man is creating a "plastic pail" out of the ocean and expecting it to continue to produce its bountiful harvest.

As you work through the physical and chemical activities in this book, try to understand them in the context of their role in the survival of sea life in any given ecosystem.

Salt water chemistry as it relates to the biotic realm is not an exact science. There are so many chemical factors involved, all of which effect each other, and so many different organisms with different tolerance ranges, that it is impossible to talk about "perfect" chemical balance. Marine chemists keep track of the water's physical and chemical changes. This information is then correlated by marine ecologists with any observed differences in species diversity and population balances.

In these exercises you will learn to measure six parameters: Salinity, Turbidity, Temperature, Suspended Solids, pH, and Light. You might ask, "What is a good turbidity level and what is a bad one?" That question cannot be answered in the absolute. The worm *Phragmatopoma sp.* which builds "worm rock," depends on turbidity, whereas many corals are very intolerant of turbid conditions. Another question you might ask is, "Are these six parameters the most important or the only important ones for marine scientists to consider?" No, they are not. Coliform bacteria counts, nitrogen levels,



dissolved oxygen readings, and many more parameters of extreme importance are monitored. It is not in the scope of this book to teach you how to monitor all important parameters. Six were chosen, however, and an attempt will be made to show how these relate to the living marine organisms.

SALINITY

Consider first, the parameter we call "salinity." You may already know the importance of this. The little sea creature in the plastic pail would have surely died if you had placed it in fresh tap water. On the other hand if the plastic pail were left standing for a time, part of the water would have evaporated leaving a saltier environment behind. Remembering that every species has a minimum and a maximum tolerance for any parameter, the poor little creature might have found his environment too salty for his continued survival. Also, the increased salinity could have lowered his tolerance for another factor such as temperature and we'd still have a funeral on our hands.

In the real world around Florida, salinity is very important in the estuaries. The grasses of our salt marshes are very susceptible to salinity changes. A change in salinity there could change the flora of that immediate area and thus alter the food chain. The distribution of mangrove species are likewise arranged to some degree by the influence of salinity: The black mangrove is more salt tolerant than the white mangrove. An increase in salinity in such areas as the Loxahatchee River has caused red mangroves to replace bald cypress. At the lower reaches of Everglades National Park, the mangrove swamp has become more saline allowing additional predators to enter this nursery ground for many marine species reducing the swamp's effective productivity. In this case, fresh water was diverted for human drinking and agricultural needs, primarily through South Florida's extensive man-made canal system. Humans, remember, have a salt tolerance also. If we continue to drain fresh water from the interior wetlands allowing further intrusion of salt water, we may find that we've put ourselves in a plastic pail.

TURBIDITY

You have already read that corals and *Phragmatopoma sp.* worms have different tolerances to turbidity. Is there more biological significance to turbidity? An emphatic YES! Egg-laying species depend upon the waters to remove CO_2 (diffusion) and to provide O_2 for absorption by the developing embryos. If these eggs are covered with a layer of silt, clay or industrial waste, embryo development is arrested. Benthic invertebrates that feed by "pumping" water have retarded growth rates or they may die *en masse.* Marine plant productivity is also decreased with less light.

Turbidity caused by a high plankton count is generally an indicator of a favorable environment. Plankton is the foundation of the marine food chain.

TEMPERATURE

Temperature is an extremely important factor in Florida. Florida is the northernmost range of such organisms as mangroves and corals. Temperature is the primary limiting factor in these cases. During cold winters, South Florida's canals rush cold water into the sea. Though the canal water has low salinity, the low temperature makes it more dense than ocean water. Thus it settles on the offshore reefs of southeastern Florida, killing many organisms with minimum salinity tolerances and those with poor tolerance for low temperatures. If water temperature increases, the water's ability to hold dissolved oxygen decreases while the metabolic rate of organisms increases. Shrimp mariculturists have discovered that by raising temperature and controlling light and adding sufficient oxygen shrimp can be brought to harvestable size sooner. Spawning of some fish species is highly influenced by

temperature. Every organism has its optimum temperature range for survival as well as temperatures which it can not tolerate.

SUSPENDED SOLIDS

Suspended particles themselves could block light, clog gills, bury coral polyps, provide building material for worms, and eventually contribute to the substrate.

Testing for suspended particles is not a complicated test. It is often used as an "indicator" for hard to detect pollutants. For example, sewage is a primary source of suspended solids. If the suspended solid count is high and the source is a sewage treatment plant, there would also be an increase in nitrogen. Nitrogen is essential to biota but it can be toxic when it is discharged in quantity by sewage plants. The presence of suspended particles alone would not cause algal blooms, but a significant increase in the number of particles should alert the investigator to look for increased nitrogen.

pH

If you have ever kept a salt water aquarium, you realize the importance of pH. The wastes of the organisms in the aquarium caused the pH to drop. In order to keep the pH up, you had to change water regularly or add a combination of sodium carbonate and sodium bicarbonate. If the pH dropped below 8.0, your fish were probably stressed and stopped eating, became less active, were susceptible to disease. Limestone gravel on the aquarium bottom, filters, and air pumps help maintain aquarium pH near 8.3.

A change in pH causes a change in the amount of oxygen that water can hold and it affects the solubility of several other chemicals. Often a change in pH changes plankton content. Just as fish waste can lower pH in aquariums, wastes produced by human populations can do the same in estuaries.

The pH can effect organisms indirectly by changing other chemical parameters and by changing organism tolerance to these parameters.

TURBIDITY, SUSPENDED SOLIDS AND LIGHT

INTRODUCTION

"Turbidity in water is caused by the presence of suspended matter, such as clay, silt, finely divided organic matter and inorganic matter, plankton and other microscopic organisms. Turbidity is an expression of the optical property that causes light to be scattered and absorbed rather than transmitted in straight lines through the sample". (Standard methods 14th Ed. 1975). The standard method for measuring turbidity has been the Jackson candle turbidimeter. For measuring lower turbidity, however, other devices were developed to give indications of the intensity of light scattered in one particular direction, predominantly at right angles to the incident light. These devices are generally called "nephelometers." Turbidities derived from the nephelometric methods are expressed as nephelometric turbidity units (NTU) and turbidities derived from visual methods are expressed as Jackson turbidity units (JTU). Various commercial nephelometers are available as well as Jackson Candle turbidimeters.

For field studies, a rough indication of the amount of suspended matter in water can be obtained with a Secchi disc. The disc is usually 20 cm in diameter. One side is white while the other may be black-white marked in quadrants.

Turbidity, suspended solids and light penetration are important to the aquatic life in that these parameters together with others, control the productivity or life processes of a wide range of plants and animals. This can occur when a large amount of silt is carried into a normally clear estuarine environment. The reduction in plant photosynthesis and possibly over a period of time the amount of dissolved oxygen available to fish and other organisms would be reduced.

It is important to note that measurements with the Secchi disc as well as other visual comparisons are only rough approximations. However, if a standard procedure is established and followed persistently a degree of precision can be reached making the measurements valuable over a period of time when compared to other relative changes.

TO THE TEACHER

Unless the class is equipped with a Jackson Candle turbidimeter or a nephelometer, sampling for turbidity should be limited to the sampling for transparency with a Secchi disc. This will give a rough indication as to the amount of suspended matter in the water. At the sampling station the student should also collect a water sample to be labeled and taken back to the lab. These samples are then filtered and dried to yield the "non-filterable" residue sometimes referred to as the *total suspended solids.* The filtrate is then placed into a beaker and slowly evaporated to dryness. The remaining "filterable" residue is often referred to as the *total dissolved solids.* It must be noted that if the beakers get too hot, some of the dissolved solids may be vaporized or decomposed.

Both suspended and dissolved solids in water cause turbidity and it is turbidity which controls the amount of light available at any given depth within the normal photo layer. Discussions may include such topics as how plankton affects turbidity, what are the advantages and disadvantages of turbidity, or what are some local sources of turbidity.

During the field readings with the Secchi disc, you may want students to do a test for color which also gives an indication of the amount of suspended and dissolved matter present. This test is done with a Florel-Ule Scale which is a comparative visual color test using standards and the color of the water over the Secchi disc. Color test kits are available from scientific supply houses.

In measuring parameters such as turbidity it is well to remember that they are affected by other parameters such as salinity, pH and temperature. Some good discussions can result from this topic and would be beneficial for the students in their understanding of all parameters and their relations to the marine environment.

TO THE STUDENT

When light enters water from the air it is reflected, refracted, absorbed, scattered and transmitted. A beam of light passing through pure H_2O has almost total transmittance. Average ocean water, however, absorbs about 54% of the blue light in the upper 10m while 10m of coastal water will absorb between 94% and 99%. This absorption limits visability with an increase in depth and even in clear ocean water, only 1% of the light energy penetrates below 100 meters. Much of the absorption and scattering is caused by the suspended material and dissolved material in water. Turbidity is a term used to express the amount of this material present in a sample of water.

In this unit you will first use a Secchi disc to measure the transparency giving a rough indication of the amount of suspended matter in the water. Then after taking a water sample, you will measure the amount of suspended solids and the amount of dissolved solids within that sample. We will then conclude with a discussion of the importance of light in the marine environment.

IN THE FIELD

Exercise I To obtain an indication of the amount of suspended matter in the water using a Secchi disc and to collect a water sample for in-the-lab determination of suspended and dissolved solids.

MATERIALS

Secchi disc
Maps
Water sampling bottles (1 for each station)
Labels or grease pencil
Field notebook

PROCEDURE

A. Secchi disc reading

1. Lower the disc into the water in a shadow during a sunny day. Reflected sunlight on the surface will obscure the vanishing point of the disc. If the current is strong, attach a weight to the underside of the disc. All readings must be made when the chain is vertical.

2. Lower the disc until it just disappears. Record the depth of the disc at this point. Raise the disc until it just reappears. Record the depth of the disc at this point. The two measurements are averaged.

This process is repeated two or three times to obtain an overall average for the Secchi disc reading. (Color of the water can be done at this time using the Florel-Ule Scale.)

B. Water sample

1. If a water sampling bottle is available, lower the bottle to the depth of the secchi disc reading and trigger. If you are using a regular screw cap plastic or other sampling bottle or jar, allow the bottle to fill below the surface.

2. Label each sample recording the location, date, time and depth at which the sample was taken.

3. Store in a cool, dark place.

BACK IN THE LAB

Exercise II - Test for Total Suspended Solids

MATERIALS

Filtering apparatus - Consisting of:
Vacuum filter flask, 750ml
One hole stopper to fit flask #7
Filter holder (funnel)
Fine filter disc (filter paper)
Petri dish
Tweezers
Water sample
Source of vacuum (aspirator)
Analytical balance
Drying oven
Large graduated cylinder

PROCEDURE

1. Determine weight of empty petri dish, with cover, after it has been marked for identification. Record.
2. Dry filter papers in a drying oven.
3. Use tweezers to select a filter from oven. Place in petri dish and cover immediately. Weigh petri dish with filter inside. Record.
4. Connect aspirator vacuum line to "arm" of filter flask. The whole filter assembly "reads" from top to bottom:
   Filter funnel
   Filter paper disc (from step 2)
   Stopper
   Filter flask
   Vacuum line
5. Determine volume (ml) of water sample using graduated cylinder. Record.
6. Agitate the sample to keep the residue in suspension. Carefully pour this into the filter funnel.
7. When whole sample is filtered, rinse the sample container with distilled water until it is optically clean, and filter this wash water. Try not to overfill the flask.
8. Remove the filter disc carefully and place in marked petri dish as before.
9. Replace petri dish in the drying oven with cover ajar.
10. Next day, determine weight of dry petri dish, filter and *non-filterable residue.* Record.
11. Subtract the weight obtained from step 2 from the weight of the non-filterable residue (total suspended solids). Record.
12. Compute the amount of non-filterable residue per liter:

$$\frac{\text{Milligrams}}{\text{Liter}} = \frac{\text{weight from step (10) x 1000}}{\text{Volume of sample in ml}}$$

If vacuum or oven are not available, a rough analysis can be made by:

1. Weigh filter paper
2. Filter known volume of sample through filter paper.
3. Allow to dry completely.
4. Re-weigh filter paper. The change in weight is the total suspended solids. Express results in mg per liter.

Exercise III - Test for Total Dissolved Solids

MATERIALS

750 ml beakers
Analytical balance
Electric hot plates

PROCEDURE

1. Weigh 3 clean, dry 750 ml beakers to the nearest 0.001gm.
2. Using the filtrate from Exercise II number 6, pour equal amounts into each beaker.
3. Slowly and carefully evaporate to dryness using the electric hot plates. Do not allow the beakers to get too hot or the dissolved solids may decompose or vaporize.
4. Once dry, place in a dustproof area and allow to cool.

5. Weigh again each 750 ml beaker and record the difference in weight from step 1 and total weights.

6. Remember the volume obtained in step 4, Exercise II should be used to calculate in mg per liter the amount of dissolved solids for this water sample. Do not use the volume which includes the wash water from Exercise II.

From Exercise II and III examine the total suspended solids and the total dissolved solids under a microscope. Describe the size and shape of the particles.

LIGHT

As you have examined the water consider the depth to which light is able to penetrate.

Consider these questions:

1. What organisms are most affected by light?
2. Are the waters near shore clearer than the water off shore?
3. What might you expect to be growing at the bottom of a deep turbid river?
4. Is turbidity always a form of pollution?
5. How do the tides affect the amount of suspended solids in the water column?
6. What are some sources of dissolved solids in the water column?
7. What color are residue and the filtrate from Exercise II? Explain. How does the color compare with the field color comparison?
8. What would be some other methods for measuring light penetration?
9. After you have analyzed a number of samples can you determine a correlation between the average depth at which the Secchi disc disappears and reappears and the mg/1 of total suspended and dissolved solids?

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SALINITY

INTRODUCTION

The term, "salinity," generally refers to the amount of dissolved salts in a quantity of sea water. Salinity is usually expressed in parts per thousand, "ppt," or grams of salt in one kilogram of sea water.

To measure salinity precisely, one would have to measure all the salts in a sample of sea water. This would be a long and tedious process. Most of the methods used to measure salinity are indirect methods which actually measure other physical or chemical properties of sea water and relate them to salinity. The amount of salt in sea water affects the water's ability to conduct an electrical current, the density of the water, and the bending of light as it passes from air into water or water into air. Other properties, such as viscosity, freezing point and osmotic pressure of water are also affected by salinity.

However, there is one property of sea water that makes it possible to measure the amount of one material and relate that to salinity. This property is that *regardless of total concentration, the proportions of all the dissolved materials in sea water are virtually constant.* These proportions hold true throughout all the oceans, seas and estuaries in the world.

One of the most direct methods to determine salinity is to evaporate a known mass of sea water to dryness and weigh the residue. There are several drawbacks to this method. Time is involved in evaporation, and weighing must be very accurate. Also, some of the solid residue may be material other than salt, and some of the salts may be volatized in the drying process.

Comparing the density of a sample of sea water with the density of pure water is another method of determining the salinity. As the salt content increases the density increases. Since density also is a function of temperature one must use both density and temperature measurements to determine salinity.

The ability of water to conduct an electric current depends upon the amount of dissolved salts and the temperature. Water is a poor conductor of electricity, but as the concentration of salts dissolved in water increases the ability to conduct an electric current also increases. Measurement of conductivity and temperature of a sample can be used to determine its salinity.

When light rays travel from one medium into another medium with a different density, they are refracted or bent. The amount of bending depends upon the density of the new medium and is expressed by a ratio called the refractive index. Since the amount of dissolved salts changes the density of water, measurement of the refractive index can be used to determine the salinity.

Pure water has a refractive index of 1.333. If the refractive index of a sample of sea water is compared to the refractive index of pure water at the same temperature the salinity can be determined.

Perhaps the most universally accepted and most accurate method used to determine salinity is by chemical analysis. As has been stated, it would be almost physically impossible to measure all of the materials dissolved in sea water every time a salinity determination is needed.

Of all the major materials dissolved in sea water, chloride ions make up approximately 55%; therefore, if one can determine the amount of chloride ions one can determine the salinity. The amount of chloride ions in a sample of sea water may be easily and accurately determined by titration with silver nitrate, using a chromate end point indicator.

TO THE TEACHER

Most of the methods used to determine the salinity of a sample of sea water are indirect methods which actually measure other physical properties and relate them to salinity. The four methods described in this unit are recommended either because they are easy to use or they are very accurate.

1. *Hydrometer* - When an accuracy of no more than 1 o/oo is needed, the hydrometer is recommended as it is easy to use and inexpensive. Plastic hydrometers are ideal for field usage but are generally not listed in scientific supply catalogues. Aquarium supply centers sometimes carry plastic hydrometers. Glass hydrometers are easily broken and care should be exercised in the field. Hydrometers are calibrated at different temperatures. The most commonly used hydrometers are based on the specific gravity of sea water at 15°C compared to pure water at 4°C. These hydrometers will have 15°/4°C on them. Some hydrometers are designated 60°/60°F. These use the specific gravity of sea water at 60°F compared to pure water at 60°F as a standard. The salinity chart in this sourcebook is for a 15°/4°C hydrometer. This method takes students through the temperature adjustment table to reinforce the density relationship.

2. *Salinometer* - The salinometer measures the specific conductivity of a sample of water as a function of salinity and temperature. This is an easy instrument to use and gives immediate results in the field. Conductivity meters are expensive, costing more than $300 for the instrument and $60 + for the probe, and they must be kept in calibration.

3. *Refractometer* - The refractometer measures the refractive index of a sample of sea water as a function of salinity. The A.O. Goldberg refractometer is temperature compensated, and the maximum error between 60°F and 100°F is 0.1%. This instrument is excellent for field work, easy to use, accurate and requires almost no maintenance; however, the cost is in the $500 range.

4. *Titration* - The titration method of determining salinity via precipitation of silver halides is a standard and very accurate method. This process involves the titration of a sample of sea water with a silver nitrate solution using a chromate end point. The students should be made thoroughly aware of the safety problems to be encountered in any exercise wherein chemicals are employed. Silver nitrate spillage must be cleaned, rinsed, and dried. Tall burets filled with titrants topple easily. The resulting mess is often wide-spread.

Overflows and spillages over the top of the buret are at eye-level or above. *Eyes must be protected at all times.*

Once silver nitrate is prepared, deterioration of the solution begins with exposure to light, evaporation and air-borne contamination. Many chemical laboratories are "rich" in HCL, H_2S and NH_4OH (NH_3) fumes. These dissolve in the opened silver nitrate to form sediments and ionic materials. Some interfere with reaching accurate end point determinations.

The end point mechanism results in the precipitation of silver chloride before the formation of red colored silver chromate. Clumps of precipitate tend to form "refuges" for the chloride ion and the silver ion interfering with a sharp determination. These clumps should be reduced by vigorous agitation periodically. The addition of 2 or 3 small plastic beads to the reacting vessel will be helpful to break clumps.

Although the pH of the sample is not made a part of this exercise, good results require a pH of 7-10. Adjust samples of pH below 7 with .1N/NaOH.

Preparation of the $AgNO_3$ titrant:

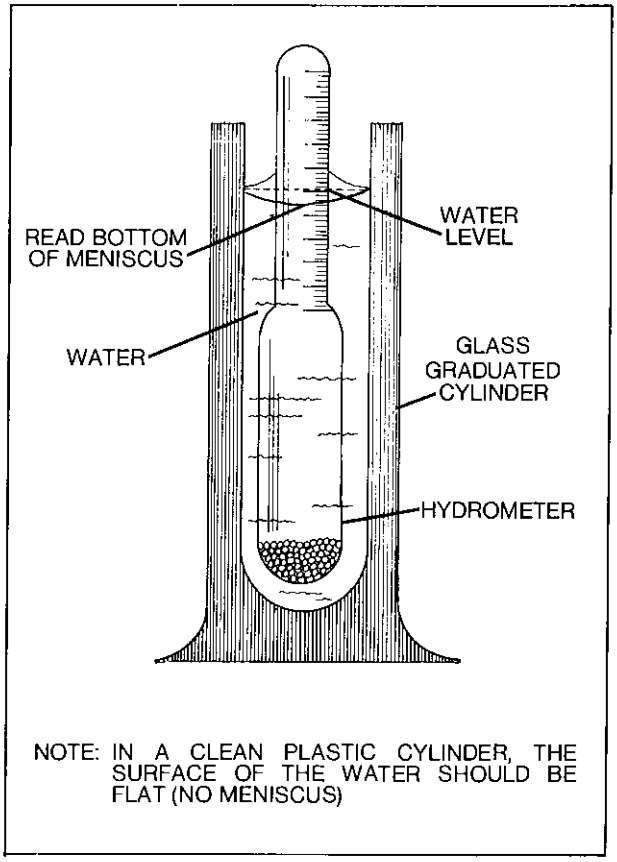
Measure 27.25 grams of crystalline $AgNO_3$. Dissolve the crystals in approximately .5L of ion-free water (distilled). Fill with more ion-free H_2O to the one liter mark.

This solution must be stored in a dark brown bottle. If several bottles are used, the contamination of one will be a lesser problem. Each buret filling requires about 50 ml of $AgNO_3$ solution. One liter will fill 20 burets (with care).

Preparation of the K_2CrO_4 indicator:

Add 5 grams of K_2 yellow crystals to 100 ml of distilled HO.

The student must be able to read the meniscus in order to correctly do this exercise.



READING THE MENISCUS IN A GRADUATED CYLINDER

TO THE STUDENT

COLLECTING THE SAMPLE

MATERIALS

Water sampler
Collecting bottles (preferably plastic)
Field notebook

Collecting bottles should be clean, and each bottle should be numbered or labeled. As each sample is collected, records should be kept on exact location of sample site, date, time of day, weather conditions, depth at which sample was taken and other information that may be used in this and other labs. Be sure the sampling method is consistent. Each sample bottle should be tightly sealed so as to prevent evaporation.

If salinity measurements are done in the field, you will need to keep the same records and include the salinity measurement. A cross reference of accuracy can be made by taking several samples to the lab and checking your field measurements with the titration method for determining salinity.

PROCEDURES FOR THE MEASUREMENT OF "SALINITY"

The hydrometer is an instrument used to determine the density of the fluid. Depending on the density of the fluid, the hydrometer will float at different levels. Most hydrometers are calibrated in terms of specific gravity, which is the ratio of the density of one substance to that of pure water at 3.98°C. In the metric system, density and specific gravity are numerically the same.

Since temperature is a factor in specific gravity, a temperature reading must be made when the hydrometer is used.

SALINITY CHART

Corresponding densities and salinities
Salinity in parts per thousand (ppt)

Hydrometer reading	TEMPERATURE OF WATER								Hydrometer reading
	55°	60°	65°	70°	75°	80°	85°	90°	
1.000	0.8	1.1	1.6	2.4	3.2	4.1	5.1	5.9	1.000
1.001	2.1	2.4	2.9	3.7	4.5	5.4	6.6	7.2	1.001
1.002	3.4	3.7	4.3	5.0	5.9	6.8	7.9	8.6	1.002
1.003	4.7	5.0	5.6	6.3	7.2	8.1	9.2	9.9	1.003
1.004	6.0	6.3	7.0	7.7	8.5	9.4	10.6	11.2	1.004
1.005	7.2	7.6	8.2	9.0	9.8	10.7	11.9	12.7	1.005
1.006	8.5	8.9	9.6	10.3	11.1	12.2	13.2	14.0	1.006
1.007	9.8	10.2	10.8	11.6	12.6	13.5	14.5	15.3	1.007
1.008	11.1	11.5	12.2	12.9	13.9	14.8	16.0	16.7	1.008
1.009	12.4	12.8	13.5	14.2	15.2	16.2	17.3	18.0	1.009
1.010	13.7	14.1	14.8	15.6	16.5	17.5	18.6	19.4	1.010
1.011	15.0	15.4	16.1	17.0	17.8	18.8	20.0	20.8	1.011
1.012	16.3	16.7	17.4	18.3	19.2	20.1	21.3	22.1	1.012
1.013	17.6	18.0	18.7	19.6	20.5	21.6	22.6	23.5	1.013
1.014	19.0	19.4	20.0	20.9	21.8	22.9	24.0	24.8	1.014
1.015	20.3	20.6	21.3	22.2	23.1	24.2	25.4	26.1	1.015
1.016	21.6	22.0	22.7	23.5	24.4	25.5	26.7	27.6	1.016
1.017	22.9	23.3	24.0	24.8	25.9	26.9	28.1	28.9	1.017
1.018	24.2	24.6	25.4	26.1	27.2	28.2	29.4	30.2	1.018
1.019	25.5	25.9	26.7	27.6	28.5	29.5	30.7	31.6	1.019
1.020	26.8	27.2	28.0	28.9	29.8	30.8	32.1	32.9	1.020
1.021	28.1	28.5	29.3	30.2	31.1	32.3	33.4	34.2	1.021
1.022	29.4	29.8	30.6	31.5	32.5	33.6	34.7	35.6	1.022
1.023	30.7	31.1	31.9	32.8	33.8	34.9	36.0	37.0	1.023
1.024	32.0	32.4	33.2	34.1	35.1	36.2	37.5	38.2	1.024
1.025	33.2	33.7	34.5	35.4	36.4	37.6	38.8	39.7	1.025
1.026	34.5	35.0	35.8	36.7	37.7	38.9	40.1	41.0	1.026
1.027	35.8	36.3	37.1	38.1	39.2	40.2	41.5	42.3	1.027
1.028	37.1	37.6	38.4	39.4	40.4	41.5	42.8		1.028
1.029	38.4	38.9	39.7	40.7	41.8	42.9			1.029
1.030	39.7	40.2	41.0	42.0					1.030

HYDROMETER METHOD

MATERIALS

Hydrometer
Thermometer
Hydrometer jar (preferably clear plastic cylinders)
Salinity conversion tables
Notebook

Float the hydrometer in your sample of sea water in the hydrometer jar or plastic cylinder. Read the specific gravity on the hydrometer at the water level. It is best to read from below the water line as the surface tension on top creates a rise around the hydrometer stem which could cause an incorrect reading. Record the specific gravity of the sample. Take the temperature of the sample and record.

Using the salinity chart determine the salinity of your sample and record.

CONDUCTIVITY METHOD (Salinometer)

MATERIALS

Salinometer
Notebook

The salinometer measures the amount of electrical current conducted by a sample of water. Since conductivity instruments are calibrated differently you should follow the instructions that come with your meter.

A few procedures should be noted, however. Before taking the salinometer into the field it should be checked for accuracy by testing a known sample. The probe should be clean and batteries in good condition. When using the probe it should not touch the bottom or other solid objects.

REFRACTOMETER METHOD

MATERIALS

Refractometer (A.O. Goldberg T/C Refractometer, Model 10419)
Notebook
Distilled water
Blotting paper
Pipette or medicine dropper

Clean the refractometer by spraying the measuring prism and cover with distilled water. (Caution - the eyepiece should not get wet. Keep all liquids below the ring). Blot the cover and prism dry. Do not rub dry.

Pipet 2 or 3 drops of sea water sample onto the prism with the cover raised and the instrument held level. Close the cover so that the sample is squeezed into a film between the cover and prism. Light finger pressure on the cover should be maintained.

Sight thru the eyepiece while holding the prism toward a light source. The borderline between the gray area above and the yellow area below crosses the scales to give the refractive index on the left and the salinity on the right.

Always clean and dry the prism after use.

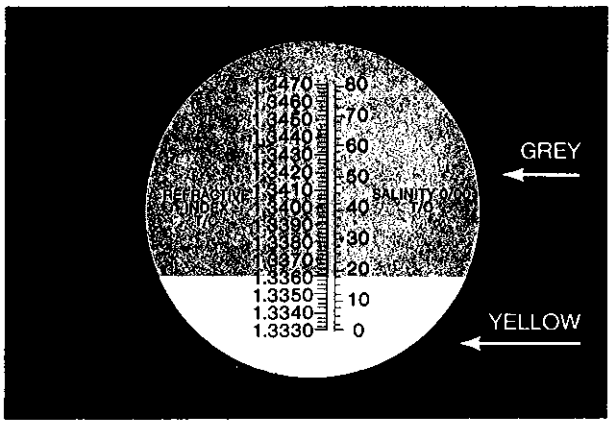


FIG. 2 SIGHT THROUGH EYEPIECE

TITRATION METHOD

Clean glassware and careful operations are essential. Begin by assuming that the table-top has been contaminated with silver nitrate by the previous class. Although $AgNO_3$ is colorless, your skin will turn black in sunlight where $AgNO_3$ is present.

Dampen a paper towel with H_2O, wipe work area with the wet towel. Then dry. Each student should wear an apron throughout the laboratory exercise. Always wear safety goggles while in the laboratory area.

MATERIALS

1–50 ml buret
1–125 ml Erlenmeyer
$AgNO_3$ titrant solution
K_2CrO_4 indicator solution
Phenophthalein
NaOH
Distilled H_2O
2–50 ml beakers
1–10 ml pipet

PROCEDURE FOR TITRATION

1. Arrange the work area for maximum usability and minimum clutter. Once titration begins, nothing should interfere.

2. Fill buret with distilled H_2O. Drain buret in short spurts. Try to adjust the stopcock so as to deliver drops and a single drop on demand. Now is the time to learn the idiosyncrasies of the stopcock. Be sure it operates without leaking.

The instructor should be informed about an inoperative buret assembly immediately.

3. Pipet 10 ml of sea water sample into the 125 ml flask. Add about 10 ml of distilled H_2O, and 2 or 3 plastic beads.

4. Put 4 - 6 drops of K_2CrO_4 into the flask. This is the indicator.

5. Fill a 50 ml beaker with $AgNO_3$.

6. Pour about 5 ml of $AgNO_3$ into the buret. Drain into the other 50 ml beaker. Pour this into sink.

7. Partially fill the buret with $AgNO_3$. Turn stopcock to fill the tip. Continue to fill the buret until there are at least 40 ml of $AgNO_3$ within the graduated scale. It is not necessary to fill the buret exactly to "0" or exactly to "50" (the top reading). This is time consuming.

8. Record the reading at the start (read meniscus). The buret either has "50" or "0" or both as the top graduation. In either case record the start and end graduations. Subtract the smaller from the larger to find the volume of $AgNO_3$ used in milliliters. Most burets can be read to the tenth of ml.

9. Drain the buret into the 125 ml flask of sea water sample in short bursts (1 ml). Touch the hanging last drop in the buret with the rim of the flask so it will run into the liquid.

10. Agitate the flask. DO NOT LOSE ANY LIQUID - to make sure, use a stopper.

11. Repeat steps 9 and 10 until the first pink-orange color appears.

12. Agitate well. The clumps of precipitate must be reduced to very small particles. The flask contents should return to the original color.

13. Add $AgNO_3$ drop by drop while agitating the flask contents sufficiently to keep the precipitate particles small. When the pink color reappears, "catch" the hanging drop. Stopper. Shake vigorously. If the pink color remains, this is the end point. Otherwise, repeat Step 13.

14. Once the end point is reached calculate the volume of $AgNO_3$ used. (See 8) THIS IS THE SALINITY. However, a correction may need to be applied - consult the table below.

Salinity Corrections (Harvey, 1963)*

Salinity, O/OO found	Correction to be applied	Salinity, O/OO found	Correction to be applied
40	−0.15	22	+0.22
38	−0.08	20	+0.22
36	−0.03	18	+0.23
34	+0.03	16	+0.23
32	+0.07	14	+0.20
30	+0.11	12	+0.19
28	+0.15	10	+0.16
26	+0.17	8	+0.15
24	+0.20		

QUESTIONS

1. When using the titration method, what additional computations must be made if one uses a sample with more or less than 10 ml of sea water?

2. In titrating, how could you "save" the experiment if too much $AgNO_3$ were accidently added to the sample.

3. Often in estuarine areas the salinity is higher near the bottom than on the top of the water. Why?

4. What is the average salinity of ocean water? Why is it sometimes much higher in shallow waters near shore?

CONCLUSIONS

Each method of determining salinity has its advantages and disadvantages. When selecting any one method over another, factors such as cost, degree of accuracy and ease of use should be considered. When using a less accurate method in the field, you should attempt to verify your results with a more accurate method in the lab.

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pH DETERMINATION OF SEA WATER

INTRODUCTION

Sea water is fresh water containing dissolved rocks, soils, minerals, and organic (plant and animal) materials. These are accumulated during the downhill rush in the creeks and rivers during that phase of the water cycle. Many of the dissolved materials then recombine through chemical reaction to form insoluble products. The remaining dissolved materials are in the form of ions, dissolved gasses and compounds.

The end product (sea water) is a mixture of solutes with the "fresh" water solvent. The final pH depends upon the excess of H^+ or OH^- ions in solution. All species of marine plants and animals have a pH tolerance. Some tolerances are "broad," i.e. pH values of 8.0 ± 1.5, and some have an extremely "narrow" pH tolerance.

Water with large numbers of water plants tends to have higher pH values than those without. Water having dissolved clays generally have lower values.

If the student has studied chemistry, much of the background material will be elemental. Otherwise, this exercise should extend and reinforce knowledge of the acid-base relationship. It should be kept in mind that your own pH meter has specific characteristics. The instructions accompanying the instrument should be followed in preference to these.

Caution! The pH of sea water has idiosyncrasies differing from water solutions containing a single solute.

The O_2, CO_2, CO_3^{-2}, HCO_3, H^+, OH^-, the metallic ions (K^+, Na^+, Ca^{+2}) and waste product ions make this sea H_2O mixture as complex as our own body fluid. Indeed, it has often been compared to blood. Thus, to seek out one relationship between only two ions (H^+, OH^-) is a gross simplification.

The negative ions of "weak" acids tend to resist large changes in pH. Hence, the term "buffer." Standardized solutions can be purchased from any chemical supply house.

Most instruments and student titrations should produce accuracies of ± 1 pH. Standard solutions prepared with saline solvents will induce a more accurate pH instrument standardization. The buffers should bracket the expected pH of 8. A single buffer at a pH of 7, 8 or 9 may be used. pH Hydrion paper covering the range of sea water is available. Try them first on saline water of known pH.

INSTRUCTIONS FOR THE STUDENT

Everyone who uses the term "pH" has an idea that it has to do with degree of acidity. Not all realize that a change of one pH number is a change of the H^+ ion concentration of pH of 10. That is: a pH of 5 has 10 times the H^+ ion concentration of a pH of 6. In a like manner a pH value of 7, although it represents "neutrality," is 1000 times as acid as a pH of 10.

What is pH then?

pH is a "short hand" way of accounting for the available acid ion (H^+). In a like manner there exists a related pOH to do the same for the available basic ion (OH).

The prevailing relationship between H^+ and OH^- is shown in two ways.

1. $H^+ + OH^- \rightarrow H_20$ and $H_20 \rightarrow H^+ OH^-$
2. The number of H^+ times the number of OH^- ions equals a constant number at a given temperature pressure and volume of water.

For ease of comparisons chemists define the standard conditions:

Temperature — Room temperature: 25°C

Pressure — One atmosphere

Volume — One liter

Thus (H^+) is a symbol representing the amount of H^+ ions in one liter at 25°C and at one atmosphere of pressure. You can now identify (OH^-).

The amount is also standardized in moles per liter. A mole of H^+ ions has a mass of one (1) gram. A mole of OH^- ions has a mass of 17 grams.

Looking again at number 2 above, write (H^+) (OH^-) = .00000000000001 $= 10^{-14}$ as the constant. Recall from algebra that $X^{-4} \cdot X^{-3} = X^{-7}$. Therefore, if the ($H^+$) can be wrtten as 10 raised to some power and (OH^-) is treated likewise, then (H^+) (OH^-) $= 10^{-14}$ can be done by adding the powers of ten. Since the "powers" system is designed for dilute solutions, the actual power of ten is usually negative (less than 1) and for economy of effort (Algebra: invert) we will change the sign on the power to plus.

Example (H^+) = 1/10 mole per liter (.1g) $= 10^{-1}$ is now called pH = 1 (the "p" is for "power") while pOH = 13 so that pConstant = 14.

Hence pH + pOH = 14.

THE PROBLEM

To find the pH of sea water.

MATERIALS NEEDED

pH indicators
pH test papers
Clean labware
pH meter
Buffers (solutions of known ph)
Distilled H_2O
Sea water samples

PROCEDURE

A. Sample collecting — The size of sample will be determined by the type and number of tests to be done. The design of meter electrode will also influence the size of sample. pH meters that are available to most students will perform very well on 50 ml. of sample. Portable meters permit pH readings *in situ* (in place).

B. Operating the meter and getting the best results.

1. Standardizing the meter:
   a. Check the meter to see that it is plugged to a "hot" outlet. (For portable meters: check the battery.)
   b. Allow warm-up period (5-10 minutes).
   c. Rinse electrodes with distilled water. Wipe dry with soft, lint-free tissue.
   d. Rinse electrode with small amount of standard buffer. (Note: a marked wash bottle containing buffer from previous standardizations is handy for this. REMEMBER: *Never* pour used buffer back).
   e. Immerse electrodes in buffer solution. Now take the temperature of buffer. If instrument has a temperature correcting dial, set to existing temperature. Otherwise, record temperature.
   f. Activate pH "Read Control"
   g. Rotate knob marked "Adjust" or "Standardize" until meter reads the correct pH.
   h. Release pH read control.
2. Reading of pH sample:
   a. Rinse electrodes with a small amount of sample.
   b. Immerse electrodes in sample.
   c. Take temperature of sample (see 1e above).
   d. Activate pH "Read Control"
   e. Record pH reading.
   f. Release pH "Read Control."
   g. Rinse electrodes with distilled H^2O and wipe.

C. *Alternate method:*

Liquid indicators and papers that show a change in color at a specific pH are available. Although there are pH Hydrion papers and others covering the expected pH of naturally occuring waters, their use is mostly confined to large variations of acidity where the decision to be made is yes or no.

Example: Is the water in the swimming pool "safe"? Yes-No. The meter is used where the specific reading is to be part of a data system for detailed analysis. Once opened, papers tend to bleach and give erratic results. Liquid indicators may cause the pH to change by interfering with the availability of the H^+ ions. Colorblind experimenters have difficulties here.

1. Tear off 2 inches (5cm) of pH paper.
2. With a clean pipet place 2-3 drops of sample on one end of pH paper.
3. Visually compare the wetted end with the chart standard accompanying the pH paper.
4. Record result.
5. Repeat steps 1-4.

DATA RECORDING

Data recording for a water sample should include the following:

Date:
Hour:
Climatic conditions:
Temperature:
Sample number:
pH:
Depth:
Collector:

QUESTIONS FOR CONSIDERATION

1. Can pH of other liquids be found?
2. If so, what body fluids can be analyzed? Blood, saliva, urine, stomach contents, and crushed plants?
3. Could one correlate pH to abundance of organisms?
4. If two industrual plants were upstream from a "fish-kill," how could a pH study identify the culprit?
5. Why should the electrodes be washed in the buffer and sample as recommended in the procedure?

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PERIODICAL

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TEACHING AID

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BIOCHEMICAL SURVEY OF AN AQUATIC ECOSYSTEM

TO THE TEACHER

Select your sampling sites prior to the field trip whether you are working from land or from boat. If sites vary physically, they will probably vary biologically. If you can trace an estuary from fresh water to the sea, you have an ideal situation. If not, then try to vary the bottom types, depth, average current, vegetation. You can reasonably expect to sample 5 sites in one school day if you do not have to travel far between sites.

Students should learn all the needed test procedures before going into the field. Rotate the students through all procedures so that all have had field experience with each test (and they won't get bored as fast).

There are many ways to make the suggested tests but meters are expensive and it usually boils down to going through chemical reactions to arrive at some colored end product which can be compared visually with a set of standards. Accuracy is usually sacrificed to affordability but test papers and other indicators suffice to make comparisons. All of these tests can be purchased in packages having easy to follow directions.

TO THE STUDENT

You are going to do a series of chemical tests on water at various field sites. You may not test for everything mentioned but all of the tests tell you something about living conditions at that site and therefore tell you something about the organisms living there.

Dissolved Oxygen (DO)—Organisms living in water have to obtain oxygen from the water. Some oxygen comes from plant metabolism. If other factors are favorable, oxygen-using organisms can survive with 4 milligrams of oxygen in a liter of water (4mg/1).

Temperature — Every organism has a minimum and maximum temperature tolerance. Often that temperature range is very broad if temperature changes occur slowly (several days) but if the temperature change is sudden the organism cannot adjust and dies. Temperature also affects how much oxygen water can hold. The higher the temperature, the less oxygen in the water.

Alkalinity — A measure of how acid or how basic a solution is. It is expressed as "pH" and may go from 1 (extremely acidic) to 14 (extremely basic) with 7 being the middle. Most marine organisms like it between 8 and 8.5.

Nitrogen — Nitrogen is vital to all life. Almost 80% of the atmosphere is nitrogen. In organisms, nitrogen is a large component of protein. When organisms decompose the protein is broken into amino acids and then to ammonia (NH_3).

NH_3 —Ammonia is essential in protein synthesis but it is also extremely poisonous. More than 0.1 mg/l spells trouble for aquatic life. Usually NH_3 is quickly converted to the nitrite ion $(NO_2)^{(-1)}$ by the organism *Nitrosomonas* sp.

$NO_2^{(-1)}$ —To convert NH_3 to the nitrite ion $(NO_2^{(-1)})$ takes a lot of oxygen and can lead to oxygen depression and fish kills. If the soil and/or water are productive the nitrite is quickly converted to the nitrate ion $(NO_3^{(-1)})$ by *Nitrobacter* sp.

$NO_3^{(-1)}$—Nitrate ion formation requires one more oxygen atom. Sometimes there is not enough nitrogen present for maximum growth and reproduction and it is a "limiting factor." Sometimes a lot of nitrogen enters the water, perhaps as fertilizer carried off the land by heavy rainfall. Nitrogen loading can cause algal blooms (rapid reproduction) and the end result could be oxygen depletion as the algae consumes oxygen for it's own metabolism. Also when the vast quantities of algae die they are processed through the nitrogen cycle removing even greater quantities of oxygen.

Phosphorus — Phosphorus in water is like nitrogen, essential to life and always combined with other elements in the form of phosphates $(PO_4^{(-3)})$. The unavailability of phosphate ion $(PO_4^{(-3)})$ can be a "limiting factor" to growth and reproduction and again, if a high concentration suddenly enters the water it can stimulate an algal bloom.

Hydrogen Sulfide — (H_2S) is the rotten egg smell sometimes associated with estuarine mud flats exposed at low tide. H_2S is a gas produced by organisms carrying on their life processes without oxygen.

PURPOSE

To do a bio-chemical survey of aquatic ecosystems.

MATERIALS

This list is general and you may not be doing all of the sampling mentioned.

Thermometer

Equipment or chemical for testing:

Salinity, Dissolved oxygen, NH_3, NO_3, NO_4, H_2S, PO_4, pH

Dredge or shovel

SITE	TEMP	SALINITY	D.O.	NH_3	NO_3	NO_4	H_2S	CURRENT/SECCHI
1								
2								
3								
4								
5								

Sieves or screen
Seine
Current meter
Water sampling bottle
Secchi disc
Containers for water to be laboratory tested
Containers for bottom sample
Containers for seine hauls
Labels for containers

PROCEDURE

1. Make data sheets like the sample shown above.
2. Make aerial view drawing of each site showing significant physical features (land contours, sea walls, water flow) and where sampling was done.
3. Write a description to accompany aerial drawing. You want to be able to return to the exact site to sample again.
4. Perform whatever tests and sampling are to be done.
5. Describe the bottom if dredging or seining was done.
6. If biological sampling is done in the field and organisms are returned to the water, record the organisms present and approximate percentage of each species to total biomass. This can be done in the laboratory.
7. Repeat the sampling at least one other time during the year to compare seasonal variations.

QUESTIONS

1. Would you expect different salinity readings at some of your sampling sites when the tide changes? Would organisms with narrow limits of tolerance to salinity live at that site? Why or why not?
2. Suppose a cattle pasture drains into a sampling site. How and why would that effect phosphate and nitrate readings?
3. Secchi disc readings tell how deep light is penetrating the water. How will the outgoing tide affect these readings?
4. If you are sampling fresh water would you expect light penetration to increase or decrease after heavy rainfall? Explain.
5. Harvesting shellfish (clams, oysters) is often stopped after heavy rainfall. Why?
6. Is seasonal temperature change at your sites going to noticeably affect a clam? If megawatt electric generators cooled by estuarine water were turned on, how would that affect the clam?
7. If you sampled again tomorrow and found a pH change of 1 point, how would the benthic organisms be affected? If that pH shift occurred over a one year period, how would the benthic organisms be affected?
8. You are a field biologist for the Florida Department of Environmental Regulation (DER) which issues permits for dredging and filling in wetlands and bodies of water. You have done this study because Acme Builders wants to fill the study area to make home sites and dredge boat channels for a connected marina. You recommend APPROVE _____ or DENY _____. Summarize your reasons for making that recommendation using the information you obtained in your study . . . remembering of course that you are not weighing alternatives or considering advantages of the project in comparison with expected environmental impact.

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MARINE MICROBIOLOGY

TO THE TEACHER

These exercises are designed to take 3 or 4 class periods. The exercise on agar digesters and luminescent bacteria can be run concurrently by extending the time to 5 days.

It is advisable to have the required media prepared ahead of time in tubes containing 5 ml each, autoclaved and stored in the refrigerator until needed. If the media are stored in test tubes with approximately 5 ml per tube, the student can melt a tube of agar and pour the required petri dish, medicine bottle, or make an agar slant. In addition, this procedure saves time when each student or team of students does not have to prepare his own media.

Marine Bacteria Agar Medium
1 liter sterile sea water
10 grams peptone
15 grams agar

Marine Yeast Agar Medium
1 liter sterile sea water
20 grams agar
23 grams dextrose
1 gram Sol-U-Pro or equivalent protein
1 gram yeast extract
100 ml Chloromycetin or Terramycin

Marine Mold Agar Medium
1 liter sterile sea water
17 grams agar
1 gram yeast extract
10 grams dextrose
100 ml Chloromycetin, Terramycin, or penicillin

Agar digester medium (3 tubes)
1 liter sterile sea water
1 gram ammonium chloride
0.5 gram galactose
15 grams agar

Marine Luminescent Bacterial Medium
20 grams peptone
10 grams glycerine
15 grams agar
1 liter sterile sea water

To prepare the media, the water should be brought to a simmer, then the agar and other ingredients added and stirred until they dissolve 5 ml of the media should be poured into test tubes and autoclaved at 15 lbs. of pressure or 120°C for 20 minutes.

Sterile test tubes and sterile sea water should also be prepared in advance and stored in a refrigerator or until needed.

For Bacteria in the Sand:
Make a trip to the beach and remove an area of sand at the low tide line approximately 2 cm. deep and 30 cm. square. Place the sand in a bucket and return to the laboratory.

For Agar Digesters:
Collect a series of samples of red algae (Rhodophyta) such as *Dasya sp., Gracilaria sp., Kalymenia sp.,* at the beach. If at all possible, the algae should be slimy to the touch. They can be stored in the refrigerator for several days prior to their use, if they are kept enclosed in a jar or a plastic bag.

For Bioluminescence:
To obtain the required bacteria culture, a fresh marine fish is kept refrigerated for a period of two days prior to the lab exercise. This is to induce maximum bacterial growth in the outer slime covering of the fish.

This exercise requires approximately 3 days. It is suggested that this exercise be done concurrently with "Agar Digesters" and "Bacteria in the Sand."

The luminescent bacterium, *Photobacterium fisheri,* can be obtained from a biological supply house and used rather than the fish culture.

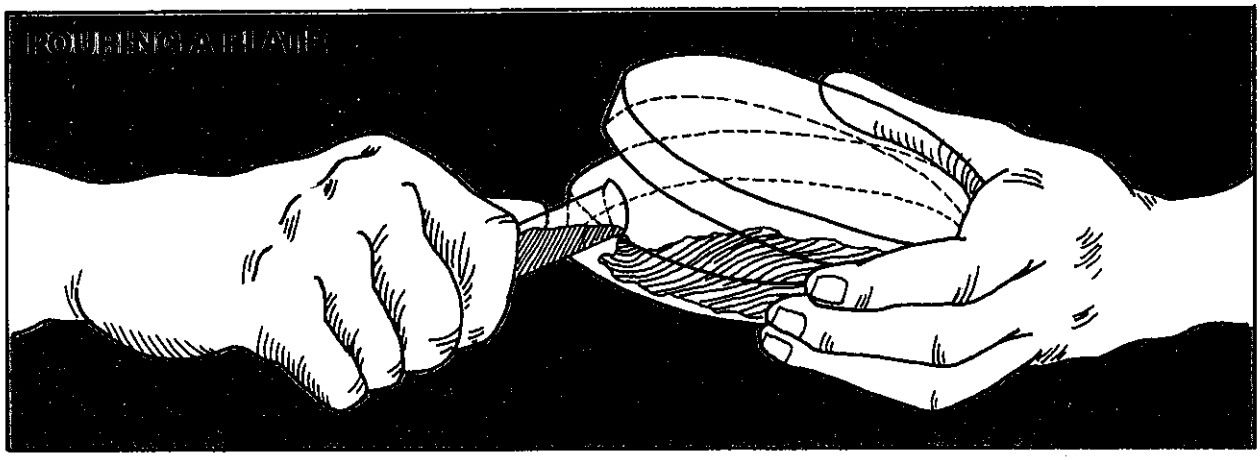
This exercise requires careful technique. The teacher should have familiarity with general bacteriology.

1. Bacteria in the Sand

TO THE STUDENT

A deserted beach appears to be a relatively sterile area, devoid of life and offering little or no protection or nutrition. If we examine the beach sand closer, we would be surprised at the abundance of life forms that exist.

Many of the life forms are microscopic bacteria, yeasts, and molds. A great many of these forms are attached to grains of sand or bits of shell. One should realize that every



time the tide changes some organisms are added and some are washed out of the beach sands.

PURPOSE

To ascertain the presence of bacteria, molds, and yeasts in beach sand.

MATERIALS

9 petri dishes
Bunsen Burner or alcohol burner
3 test tubes containing 5 ml each of marine bacteria Agar medium
3 test tubes containing 5 ml each of marine yeast Agar medium
3 test tubes containing 5 ml each of marine mold Agar medium
9 slides and cover slips
Dissecting microscope or hand lens
Compound Microscope
Crystal violet stain 2%
Sterile sea water
Inoculation loop or sterile "Q-Tips"
Grease pencil
Sterile test tubes

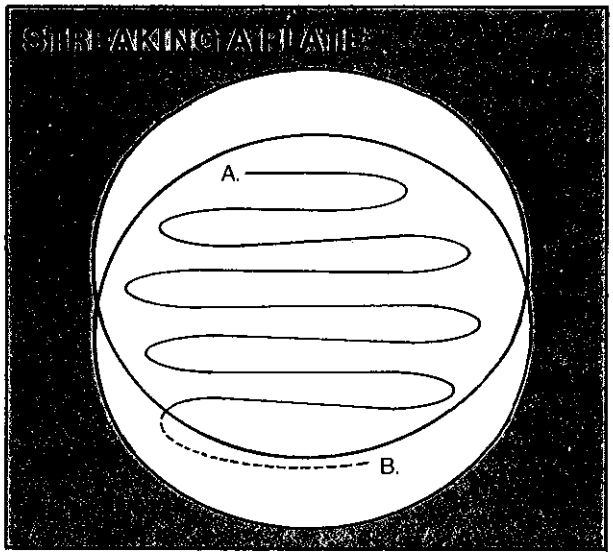
PROCEDURES

1. Prepare a minimum of 3 petri dishes of each type of medium by the following method:

After melting a test tube of the desired medium, the tube should be held in the right hand with the petri dish lid opened by the left hand. The lid should be opened only enough to allow the mouth of the tube to be inserted and its contents emptied into the petri dish and then closed as rapidly as possible to prevent contamination.

The entire process should be carried out in a smooth flowing motion rather than a hurried jerking motion.

Allow the media to harden.



2. Mark the plates with a grease pencil to denote the type of medium used i.e. M=mold; Y=yeast; and B=Bacteria.

3. Add ⅓ tube of the sand sample to ⅓ tube of sterile sea water, stopper, and agitate thoroughly.

4. Permit the sand to settle in the tube, then decant 1 ml of the liquid into a sterile tube for your use.

5. The inoculation loop should be passed through the blue portion of a flame *before* and *after* each use.

The mouth of a test tube is to be flamed before and after each opening of the tube and the tube held at a downward angle when open to prevent contamination falling into the tube from air currents.

Using your inoculation loop or "Q-Tip", streak two of the three petri dishes from each type or agar as illustrated (one dish from each group is to be kept as a control).

Begin at position "a" with the inoculating loop and lightly draw the loop across the agar surface in the illustrated pattern, being careful not to break the surface of the agar. On finishing at position "b" the plate should be closed immediately and the inoculation loop flamed.

6. The dishes of inoculated agar plates are to be kept in a warm, humid atmosphere for the next 4 days, preferably in an incubator with a dish of water to provide humidity.

7. The following observations are to be made each day and

recorded on the chart provided:

a. number of colonies present
b. color, shape, and size of each colony
c. growth pattern for each colony

From each type of growth on each plate, remove a sample of the colony with the inoculation loop and mix with a drop of sterile sea water on a clean slide. Cover with a cover glass and observe under both low and high power of the microscope.

8. After the initial observations, each slide is to be stained with crystal violet 2% stain by: (a) removing the cover slip; (b) pass the slide gently through a flame 3 or 4 times or until dry with the wet side up; (c) when the slide cools to room temperature, cover the slide with 2 or 3 drops of the stain; (d) allow the stain to remain on the slide for 30 seconds, then pour off the excess; (e) the slide should now be rinsed in a beaker of clean water by immersing gently; (f) the water left on the slide is removed by blotting gently with a paper towel - DO NOT RUB.

9. The slides are now ready for observation under the microscope.

10. List and illustrate the organisms observed at this time. Identify each according to the medium it was grown on.

QUESTIONS FOR CONSIDERATION

1. Did the microorganisms occur in the sea water or the sand?
2. Why would these organisms exist in the sand?
3. Why is each medium of different composition?
4. What function do these organisms have in the marine food chain?
5. At what point(s) in the procedure is contamination most likely to occur?

2. Agar Digesters

TO THE STUDENT

In the red algae, the agar material is a carbohydrate that aids in building the plant cell wall. Since bacteria are present everywhere in the marine environment, it would be expected bacteria will be found on those algae which are used to produce agar commercially.

PURPOSE

To detect and demonstrate the presence of bacteria which use agar as food and to illustrate the variety of marine organisms which furnish support for bacterial life forms.

MATERIALS NEEDED

Bunsen Burner or Alcohol lamp
Red algae
3 Sterile petri dishes
Sterile Knife
Inoculating loop
Test tube
Iodine, 5% aqueous
Sterile sea water
3 test tubes containing 5 ml each of agar digester medium

PROCEDURE

a. Pour the test tubes of prepared media into the sterile petri dishes and allow them to harden.

b. Select a portion of red algae with as large a surface area as possible.

c. Using a sterilized knife, scrape a portion of slime from the algae surface.

d. Rinse the slime from the knife into a test tube containing 10 ml of sterile sea water.

e. Shake the tube to disperse the slime evenly into suspension.

f. Using the inoculating loop, streak several drops of the suspension over the surface of two of the petri dishes. (The third dish becomes the control plate to demonstrate lack of contamination.) Be careful not to break the surface of the agar.

g. Incubate the petri dishes at a relatively constant temperature and humidity.

ORGANIZED DATA RECORDING

Observe each petri dish daily and count the number of colonies present. Record the type of colonial growth and the occurence of pits and depressions. Record the size change per colony on a daily basis. Check the control plate daily.

After a number of pits and depressions have developed, one of the two inoculated dishes should be flooded with iodine solution. After one minute pour off the excess iodine and record your observations. Repeat this two or three days later with the second dish and record your observations.

Analyze the iodine test on the petri dish for color production, effect on the medium by the bacteria, and other uses.

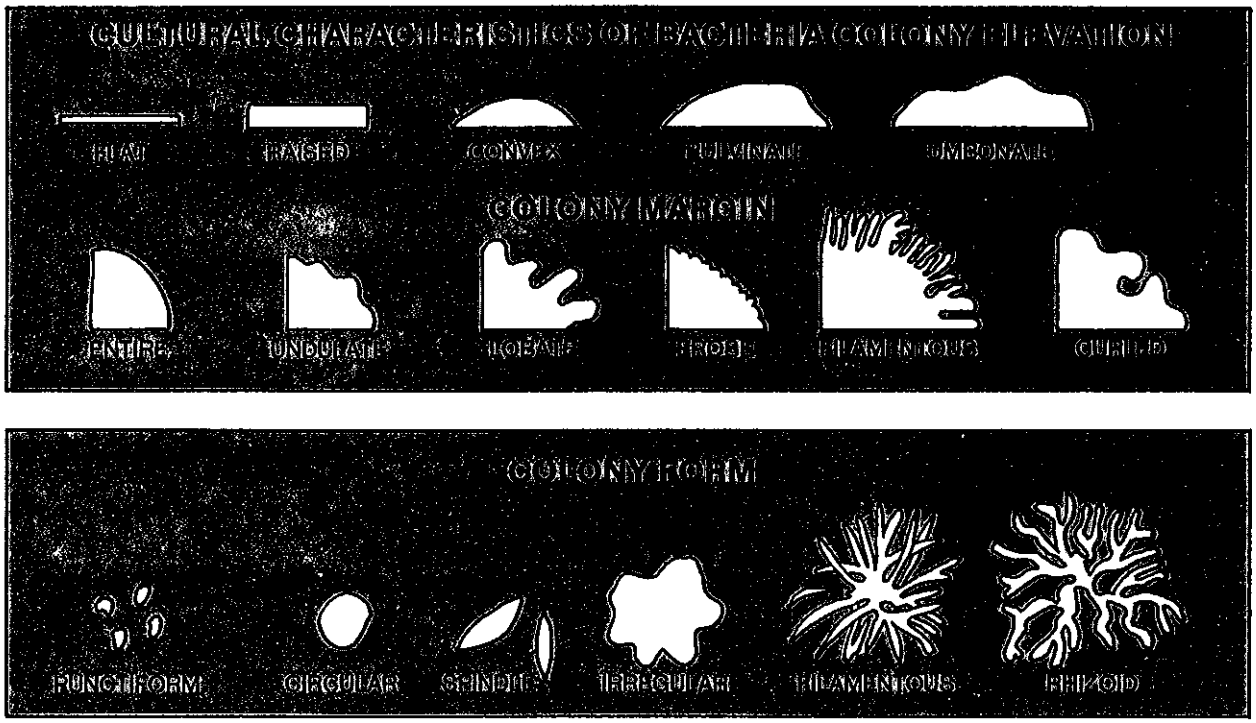
QUESTIONS FOR CONSIDERATION

1. Is the agar in depressions consumed?
2. What is the source of carbon for bacteria in the sea?
3. If bacterial growth occurs on the control plate, account for this.
4. Why are red algae selected as the source for the bacteria?

3. Bioluminescence

TO THE STUDENT

Some organisms possess a spectacular energy-display mechanism, luminescence. "Fireflies" or "lightning bugs" are the most familiar life forms to exhibit this characteristic.



It is less well-known that bacteria also possess the ability to produce light.

Bioluminescence is produced by the action of an enzyme (luciferase) on a substance known as luciferin. The reaction produces visible light. "The light emission of higher organisms may have a definite purpose . . . for attracting the sexes, as a lure for food or warning signal, perhaps for illumination . . . but it is practically impossible to assign a function of value to luminous bacteria in the above sense". (Harvey, 1952)

The smallest flash of light is seen immediately at night. This explains the numerous times a luminous wake is observed behind a boat or the wonder at the glow of the castnet used at night. This is due to the number of dinoflagellates and comb jellies that emit light when disturbed.

It has been found that bioluminescence is 100% efficient in light production. One molecule of luciferin consumed or burned produces one unit of light. This is described as "cold light" as no heat is produced. All energy is thus converted into light. (McElroy and Seliger, 1962)

Lucifren + Oxygen = Oxy − Luciferin + Water (+ light) (Russell and Yonge, 1963)

The number of organisms that exhibit bioluminescence is legion. They include bacteria, fungi, radiolarians, dinoflagellates, sponges, jellyfish, seapanseys, comb jellies, marine worms, squids, crustaceans, clams and snails.

MATERIALS

Sterile petri dishes
Sterile test tubes
Inoculating loop or "Q-Tip"
3 test tubes containing 5 ml each of marine luminescent bacteria medium
Bunsen burner or alcohol burner

PROCEDURE

a. Prepare 3 petri dishes of medium. Use one dish as a control plate.

b. Scrape a layer of surface slime from the side of the fish, using a sterile instrument.

c. Place the scrapings on a sterile test tube containing 10 ml of sterile sea water

d. Agitate the test tube to mix thoroughly.

e. Use the inoculating loop or "Q-Tip" to streak two petri dishes with the solution.

f. Incubate all three petri dishes in a cool, dark place (not the refrigerator) and observe daily in a dark room.

g. When luminescent colonies of bacteria are found, use the inoculating loop to scrape the colony from the surface of the agar.

h. Transfer the bacteria to a sterile test tube containing 5 ml of sterile sea water and observe in the dark.

i. Shake the tube and repeat observations.

ORGANIZED DATA RECORDING

Observe and record daily:
Number of colonies
Size of colonies
Form of colonies
Time of appearance of each colony
Amount of luminescence per colony
Amount of luminescence in test tube before and after shaking

QUESTIONS FOR CONSIDERATION

1. Why does luminescence exist in the plant and animal world?
2. What conclusion does the refrigeration of the fish and incubation of the petri dishes lead you to make?
3. What relationship exists between the fish and bacteria?
4. How common is luminescence among the plant and animal phyla?
5. Why do some deep sea fishes possess bioluminescence?

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TAXONOMY OF MARINE ORGANISMS

A general exercise to introduce the major groups of marine animals and their characteristics.

INTRODUCTION

Taxonomy is the science of the classification of all living things. We now recognize Linnaeus' "Systema Naturae" (1758) as the beginning of an organized scientific effort to name the plants and animals of the world. Do not become discouraged with the variety of methods used to identify organisms — ideas have changed as science has progressed. Some organisms are very difficult to classify — they simply do not seem to fit into any scheme of classification. Remember that any modern system of taxonomy tells us much more than just the name of the animal. Its relationship to all other animals (like a family tree) and the progressive development or degeneration over millions of years can often be shown by proper classification. Remember, too, that we like to know the names of all things. Most of us are basically curious; we want to know *who* as well as *why*. If we go to a party and there are too many strange people we feel ill at ease until we are introduced. We hope to introduce you to the animals of the sea and the seashore. Since there are over 500,000 animals which have been named we can only become acquainted with a few from each major group. In science we use the binomial system of classification. For example the common blue crab is called *Callinectes sapidus* Rathbun. *Callinectes* is the genus, *sapidus*, the species, and Rathbun means that Dr. Mary J. Rathbun, who was an authority on crabs, first described this species scientifically. The word species means that the crabs of this group have the same characteristics and that they can mate and produce fertile offspring.

TO THE TEACHER

In the audio-visual list at the end of this exercise, we have listed the titles of a few films. We can think of no more appropriate time to use any available films related to marine animals. The diversity of marine life and many ecological principles are more beautifully illustrated by films than by any other method. The purpose of this laboratory exercise is to introduce the student to the major marine phyla. Detailed study of certain species will come later. This is just a wide-angle overview of the marine environment. (See the Summary of Marine Animal Phyla appended to this exercise.) The length of time spent on this lab depends on the nature of the course — whether there is only a unit on Marine Biology or an entire year. It is suggested that students start with the larger forms of the invertebrates.

One of the most interesting methods of introducing the student to the various phyla of marine organisms is thru the salt water aquarium. A number of small aquaria can be used to teach not only phylogenetic grouping but habitat selection, feeding habits, and many other methods of grouping organisms.

In many areas, a collecting permit will be necessary. The teacher should contact the proper authorities before taking a collecting field trip.

TO THE STUDENT

At low tide specimens can simply be picked up from the beach. If the water is not too deep for wading, nets made of cotton mesh or nylon mesh can be used. Diving masks or glass-bottom buckets are a great help in locating specimens. The use of SCUBA by students on field trips is not recommended. A certified, professional dive master should always be present on any diving field trip and have total authority in all diving aspects of the trip.

Most specimens collected can be observed in the field and released. *Over-collecting must be avoided.* If specimens are to be taken back to the classroom aquaria several questions should be answered first: Is there enough room in the aquarium for the new specimens? Will we be able to feed the new specimens? (some animals are only filter feeders); Is there enough light to permit plant growth? Do we have enough time and aeration to get the specimens back to the classroom? If there is not a salt water aquarium and if you cannot keep the specimen alive, the specimen may be preserved in 70% ethyl alcohol in sea water. Invertebrates are better preserved in formalin solution (10%) and later washed and transferred to 70% ethyl alcohol.

Every student should have a field notebook to record all details of the field trip; the date, exact location of the collecting area, weather and tide information. Information regarding wave height, water temperature, and the direction of the wind at the time of the field trip and for several days previous is helpful. Every specimen should be carefully labeled.

The Smithsonian Institute in Washington, D.C., now has over 30 million named biological specimens. With the interest in the oceans increasing so rapidly, so many specimens were being sent to the Institution that an Oceanographic Sorting Center was created. To date, over eight million specimens have been sorted and over three million specimens have been shipped to specialists for detailed study. On a small scale, the same procedure is being followed in this lab.

THE PROBLEM UNDER INVESTIGATION

The problem is to separate the animals collected on the field trip into the proper phyla. To assist in this work there is appended a summary of the Phyla of Marine Animals. It is not complete, for one could spend a lifetime studying a small group of one phylum. Listed are common names of some representative animals belonging to each phylum. As common names vary considerably, the Phylum Summary should be revised to conform with indigenous common names.

MATERIALS

The materials needed and the techniques of collecting from the various marine environments are discussed elsewhere in the source book. A deep-freezer is invaluable in our taxonomic work. For example, a fish can be placed on a piece of cardboard and the fins spread out and pinned. The specimen is then frozen. If glycerine is rubbed on the specimen, the frosting which obscures details can be reduced. A very large study collection can be maintained in a deep-freezer. It is particularly good for larger specimens and is ideal when laboratory time is at a premium.

PROCEDURE

The classification of the animals will be based on their external anatomy. Under study will be the larger animals which can be seen with the unaided eye. Microscopic forms will be considered in other labs. Roughly sort the specimens according to features which are alike or dissimilar. After this rough sorting is done, see how many animals can be sorted to phylum. Someone may have an extensive collection of seashells; another student may know the human resource material first — individual knowledge. After this rough sorting, it is advisable to divide the class into groups of "specialists", — shell specialists, fish specialists, "worm" specialists, etc., who then start the classification of the specimens in detail.

ORGANIZED DATA RECORDING

Every identified specimen should be properly labeled. A suggested label form is shown below. A 3" x 5" index card should also be made for each species and maintained in a separate card file. It is hoped that in time this biological data may be correlated on a state-wide basis.

QUESTIONS FOR CONSIDERATION

1. What do we mean by the word phylum?
2. One job of a taxonomist is to name things. What other information can he give us?
3. What is a dichotomous key?
4. Most animals are classified according to their external characteristics. By what other methods could animals be classified?
5. Why is it that color is not a good basis for classification?
6. How would you define species?

Suggested Label Form

Technical name -
Common name -
Family -
Habitat -
Locale -
Description -

Date -
Collector -

A SUMMARY OF MARINE ANIMAL PHYLA

The only time that anything is simple or easy is either when we do not know anything about the subject or when someone else does the work. This is certainly true in the classification of animals. With over a million known species, it is impossible to know more than a very few. The study of one or two species may be the work of a lifetime. There are many methods of classifying animals. Used here is a system combining those found in Otto and Towle, *Modern Biology;* Dawson, *Marine Botany;* and Barnes, *Invertebrate Zoology.*

Kingdom: Monera

Phylum: Schizomycophyta
Bacteria and related organisms — cells lack nuclear membranes — found through out the marine environment — most numerous in coastal waters — function in nitrogen fixation, sulfate reduction, marine fouling, decomposition and decay and as pathogens

Phylum: Cyanophyta
Blue-greens algae — cells lack nuclear membranes — have chlorophyll but not organized into plastids — found throughout the marine environment — most numerous in benthic environment.

Kingdom: Protista

Simple, unicellular, colonial or multicellular organisms without true tissues — not distinguishable as plant or animal.

Phylum: Chlorophyta
Green alage — cells contain organized nucleus and plastids with food stored as starch; cell walls have cellulose; some calcified forms; generally have a vivid, green color due to the presence of both chlorophyll *a* and chlorophyll *b*; rarely dominant in phytoplankton; most are benthic forms, and are primarily macroscopic

Phylum: Chrysophyta
Yellow-green algae and diatoms — cells contain organized nucleus and plastids with food usually stored as an oil. There is no chlorphyll *b* present. Most are planktonic although there are many benthic diatoms; many with cell walls containing silicone (diatoms) or calcium carbonate (coccoliths).

Phylum: Pyrrophyta
Golden-brown algae, dinoflagellates — cells contain organized nucleus and plastids with food stored as a starch or oil; have flagella; abundant in plankton; many are luminescent; all are unicellular; some species responsible for "red tide" blooms.

Phylum: Phaeophyta
Brown algae — cells well organized with plastids; Chlorophyll *a* and chlorophyll *c* present but masked by the pigment fucoxanthin to produce brown appearance; mostly multicellular and macroscopic; almost all are attached, benthic forms.

Phylum: Rhodophyta
Red algae — cells well organized with plastids; the pigment phycoerythrin gives red appearance although other pigments are present; most are multicellular and macroscopic; most numerous of algal groups; range from shallow to very deep water; most numerous in temperate and tropical waters; many calcified, coralline forms.

Phylum: Mycophyta
True Fungi — do not have chlorophyll; all are parasitic or saprophytic; role in decomposition of organic material; many are pathogenic in animals and plants; from shallow to very deep water.

Phylum: Sarcodina
Protozoans with pseudopodia; ameboid-like form; some secrete a calcium carbonate shell, (foraminiferans); some form a siliceous skeleton, (radiolarians); planktonic and benthic dwellers.

Phylum: Mastigophora
Protozoans with flagella; many are symbiotic or parasitic

Phylum: Ciliophora
Protozoans possessing cilia used for locomotion; numerous in estuarine environment; the family, Tintinnidae is well represented in marine plankton throughout the world.

Phylum: Sporozoa
Protozoans possessing no structures for locomotion; all are parasitic; usually include a spore stage in lifecycle.

Kingdom: Metaphyta
Autotrophic, multicellular plants

Phylum: Tracheophyta
Plants with vascular tissue

Class: Angiospermae
Flowering plants

Subclass: Monocotyledonae
embryo with one cotyledon; leaves parallel-veined; submerged and emergent plants found in coastal waters

Subclass: Dicotyledonae
Embryo with two cotyledons; leaves with netted veins; mangrove associations along tropical and subtropical coastlines

Kingdom: Metazoa

Phylum: Porifera
Sponges — body cyclindrical branching or irregular; skeleton of spicules or spongin or both; body has many small pores and one or more large openings.

Class: Calcispongiae
Calcium carbonate spicules; asconoid, syconoid and lenconoid, canals; simple limy sponges

Class: Hyalospongiae
Siliceous spicules (6-rayed); syconoid canals and leuconoid canals

Class: Demospongiae
Siliceous spicules (not 6-rayed) and/or spongin; leuconoid canals

Phylum: Coelenterata (Cnidaria)
Body sack-like; interior of body functions as a digestive sac; luminescence common; all have stinging cells (cnidoblasts); live in a variety of habitats.

Class: Hydrozoa (hydroids)
Hydra. Solitary or colonial polyp and/or medusa stage present. Portugese man-of-war.

Class: Scyphozoa (jellyfish)
True jellyfish; medusa dominant; polyp inconspicuous or absent.

Class: Anthozoa ("flower-animal")
Polyp stage only; marine only

Subclass: Soantharia (Hexacorallia)
Sea anemone; hard coral - 6 (or some multiple of 6) septa, simple tentacles

Subclass: Alcyonaria (Octocorallia)
Soft coral; sea pansies — 8 septa; pinnate tentacles

Phylum: Ctenophora
Have 8 comb plate rows (ctenes); colloblasts; triploblastic; all are marine.

Phylum: Platyhelminthes
Dorsoventrally flattened; flame cells; not segmented.

Class: Turbellaria
Free-living; gut present; planarians.

Class: Trematoda
Ecto and endo-parasites; gut present; flukes

Class: Cestoda
Endo-parasites; gut absent; tape worms.

Phylum: Rhynchocoela
Ribbon worms; cilia absent (exception—flame bulbs)

Phylum: Rotifera
Ciliated crown; wheel animals.

Phylum: Gastrotricha
Cuticle with scales or spines.

Phylum: Nematoda
Unsegmented roundworms; cilia absent.

Phylum: Mollusca (Soft-Bodies)
Body composed of head foot, visceral hump covered by mantle that secretes shell.

Class: Monoplacophora (Single plate bearer)
Ancient, segmented limpet-like shell internal metamerism.
Class: Amphineura (Both Side Nerves)
Body dorsoventrally flattened; head reduced, food highly developed.
Class: Scaphopoda (Boat Foot)
Tooth shells; hollow tentacles; head reduced.
Class: Gastropoda (Stomach Foot)
Body twisted; shell spiral (when present); snails.
Class: Pelecypoda (Hatchet Foot)
Bivalves; marine and fresh water; filter feeders; clams, oyster, etc.
Class: Cephalopoda (Head Foot)
Shell chambered or reduced, internal, external as in Nautilus, or absent; well developed head and eyes, ganglia fused and centralized; octopus, squid.

Phylum: Annelida (Little Ring)
Segmented worms; setae for locomotion
Class: Polychaeta
Parapodia with many setae; dioecious; mostly marine
Class: Hirudinea
Parapodia and chaetae absent; monoecious; mostly fresh water; some marine leeches.

Phylum: Arthropoda
Chitinous exoskeleton; jointed appendages.
Subphylum: Trilobitomorpha
Extinct marine arthropods; trilobites.
Subphylum: Mandibulata
Mandibles (jaws) and antennae present.
Class: Crustacea
2 pairs antennae; most are marine.
Subclass: Brachiopoda
Mostly fresh water, appendages used as gills; fairy shrimp, brine shrimp.
Subclass: Ostracoda
Ostracods; body enclosed in a bivalved carapace.
Subclass: Copepoda
Copepodas; planktonic and bethic; filter feeders, and parasites.
Subclass: Branchiura
Fish lice; ectoparasites on fish
Subclass: Cirripedia
Adults sessile; many parasitic, barnacles.
Subclass: Malacostraca
Usually form calcareous plates around body 19 segments; 5 head, 8 thoracic, 6 abdominal.
Order: Stomatopoda
Mantis shrimps; second appendage developed for raptorial feeding; bethic.
Order: Isopoda
Body dorso-ventrally flattened; Wharf Roach.
Order: Amphipoda
Body laterally flattened; Amphipods
Order: Euphausidacea
Krill-shrimp; pelagic; filter feeders
Order: Decapoda (Ten Foot)
3 pr maxillipeds; 5 pr "walking" legs
Sub order: Natantia
Body laterally flattened; shrimp.
Sub order: Reptantia
Body dorso-ventrally flattened; lobsters and crabs.
Class: Insecta
Very few marine species; body in three parts; three pairs of legs.
Subphylum: Chelicerata
Chelicerae present (two pincers used for eating).
Class: Merostomata
Body divided into cephalothorax and abdomen; 5 or 6 pairs of abdominal appendages; horseshoe crab.
Class: Pycnogonida
Abdomen reduced; sea spiders
Phylum: Tardigrada
2 pair of unjointed appendages; water bears.
Phylum: Sipunculida
Unsegmented worms; body divided into slender introvert and wider trunk; peanut worms.
Phylum: Echinodermata (Spiny-skinned)
Radially symmetrical water vascular system; no head; endoskeleton of calcareous ossicles or plates.
Class: Stellaroidea (Star-like)
Subclass: Asteroidea
Arms not sharply marked off from central disc; sea stars
Subclass: Ophiuroidea
Arms sharply marked off from central disc; brittle stars.
Class: Echinoida (Spiny-like)
Globular; without arms; with skeleton (test) composed of fused plates; sea urchins.
Class: Holothuroidea (Plant-animal like)
Sausage shaped; without arms; skeleton internal and reduced; sea cucumbers.
Class: Crinoidea (Lily-like)
Sea lillies; feather stars; usually sessile as adults; no spines.
Phylum: Chaetognatha
All are marine; almost all are planktonic: arrow worms.
Phylum: Hemichordata
Worm-like body divided into proboscis, collar and trunk; common mud flat animal; acorn worms.
Phylum: Chordata
With gill slits or remnants, embryo with notochord (a dorsal hollow nerve cord).
Sybphylum: Urochordata
Notochord restricted to tail of embryo; sessile to pelagic; sea squirts and relatives.
Subphylum: Vertebrata
Coelomate, segmented notochord in embryo only; nerve cord well developed.

Superclass: Cyclostamata
Lampreys and hagfishes; round mouth; lack jaws and paired appendages.
Superclass: Gnathostomata
Having jaws and paired appendages.
Class: Chondrichthyes
Sharks, rays and chimeras
Class: Osteichthyes
Bony fish.
Class: Amphibia
Frogs
Class: Reptilia
Snakes and turtles
Class: Aves
Birds
Class: Mammalia
Mammals

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AUDIO-VISUAL LIST

There are many excellent films on marine life. A few are:

The Sea — Encyclopedia Britannica Films. (26 min. color) The infinite diversity of the structure and function of marine organism is beautifully shown.

Beach and Sea Animals — (2nd) edition Encyclopedia Films (12 min. color). Shallow water animals illustrated by some unusual underwater photography.

Adaptive Radiation and The Mollusks — Encyclopedia Britannia Films. (18 min. color) This covers only the mollusks.

Films on Oceanography — (1966) 3rd edition, Publication C4 National Oceanographic Data Center, Washington D.C. Many of the films listed in this catalog are free.

STUDY OF A CRUSTACEA —PINK SHRIMP

INTRODUCTION

The pink shrimp, *Penaeus duorarum*, is a very common marine crustacean found seasonally in bays and estuaries and year-round in the grass beds of the Gulf of Mexico. The pink shrimp is a commercially important species both as food and as bait.

TO THE TEACHER

Through the study of both live and preserved specimens, the students will gain a knowledge of the major anatomical and behavioral adaptations of the pink shrimp. The lab can be completed in one 55-minute period or extended over several days with a comparison of the anatomy and behavior of the pink shrimp with other decapod crustaceans.

TO THE STUDENT

After completing this lab, you should be able to:

1. Match the major external anatomical features of the pink shrimp with their functions.
2. Label a drawing of the pink shrimp showing all the major anatomical features.
3. Observe and describe several behaviors of the live shrimp and indicate how these behaviors help the shrimp survive.
4. Describe the type of environment for which the shrimp is best adapted.

PURPOSE

To study the anatomical and behavioral adaptations which help a pink shrimp survive in its environment.

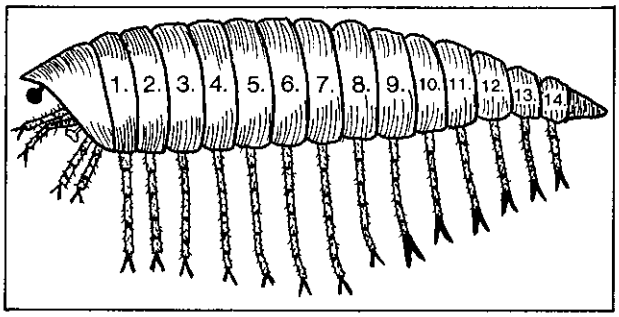
MATERIALS

Live and preserved shrimp
Aquarium with sea water and at least two inches of sandy substrate.
Dissecting pan
Dissecting kits
Dissecting microscope
Small hand net
Fine-grained fish food

PROCEDURE

EXTERNAL ANATOMY

The pink shrimp belongs to the subclass of crustaceans known as *Malacostraca*. The subclass contains almost three quarters of all known species of crustaceans.



The body of a typical malacostracan is composed of a head followed by 14 body segments. The first eight segments form the thorax and the last six the abdomen. The body of your shrimp has been modified by natural selection and no longer fits this plan completely. How does the pink shrimp's body differ from the typical malacostracan plan? (1)_____

_____

_____

In some crustaceans such as your shrimp, the head and thorax are fused and covered by a single shell known as a *carapace*. The fused head and thorax is often referred to as the *cephalothorax*. On the pink shrimp, the cephalothorax is laterally compressed. On other crustaceans, such as crabs, the cephalothorax is dorso-ventrally flattened.

At the front (anterior) end of the cephalothorax is a sharp pointed projection known as a *rostrum*. What might be the function of this structure? (2)_____

_____

Does it offer any protection for the head of the shrimp? (3)_____

Note that there are three sets of appendages associated with the head region. Find the stalked eyes, what is their function? (4)_____

_____

The other two sets of appendages are used to sense the shrimp's environment. The long pair are known as antennae and the double short pair are known as antennules. How might the function of these appendages differ? (5)_____

_____

The longer antennae function as long range sense organs to detect other ogranisms in the shrimp's environment. The antennules are short range detectors. They sense food and other stimuli close to the shrimp's head.

Eleven other appendages are associated with the cephalothorax. The last five pairs of thoractic appendages are called walking legs. What do you suppose they are used for? (6)_____

If you examine the legs closely you will notice that the last segments differ. In the space below, draw one of each type of leg and label it with a number. The most anterior pair is one and the most posterior is five. (7)

As you have noticed, some of the walking legs have pincers on them. What might be the function of those structures? (8)_____

Cut away a section of the carapace just above (dorsal to) the legs. Notice the soft, spongy, feather-like structures attached to the top of the legs. These are the gills. What are they used for? (9)_____

When the legs move, so do the gills. How does this help the shrimp get oxygen from its environment? (10)_____

Anterior to the walking legs are the feeding appendages. These consist of the three pairs of large leg-like appendages known as maxillipeds, the two smaller pairs of leg-like appendages known as maxillae and a pair of hard rough appendages known as mandibles.

The maxillipeds take material picked up by the walking legs and separate food particles from sand, shell or other non-edible material. They then cut the food up into small pieces. The maxillae cut the food up even smaller and the mandibles grind it into tiny particles which are then swallowed.

Posterior to the cephalothorax are the six segments that make up the abdomen. Why are most people interested in this part of the shrimp? (11)_____

The appendages attached to the first five abdominal segments are known as swimmerettes. What is most probably their function? (12)_____

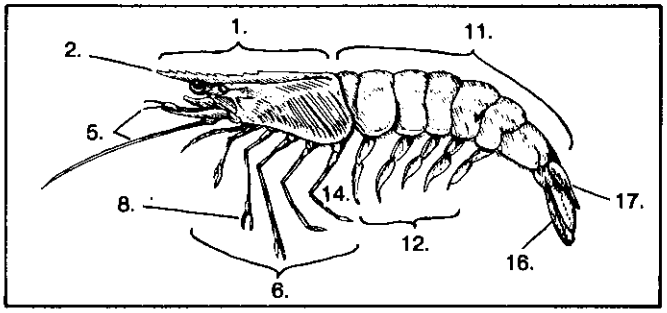
Are all swimmerettes alike? (13)_____. If all your swimmerettes are alike, find a shrimp on which some are different. Which pairs are different? (14)_____

The shrimp with modified swimmerettes are males. What do you suppose these first two pairs are used for? (15)_____

Attached to the last abdominal segment is a tail fan. It is composed of a modified pair of swimmerettes called *uropods* and a sharp terminal segment, the *telson.* What might be the function of the uropods? (16)_____

The *telson?* (17)_____

Using what you have learned, label the following diagram.



1._____
2._____
3._____
4._____
5._____
6._____
7._____
8._____
9._____
10._____
11._____
12._____
13._____
14._____
15._____
16._____
17._____

Now that you know something about the parts of a shrimp, let's see what you can learn about its behavior.

BEHAVOIR

First, locate the live shrimp in the aquarium. What is the shrimp's normal method of locomotion? (18)_____

Can the shrimp swim both forward and backward using their swimmerettes? (19)_____

What appendages do the shrimp use to escape capture? (20)_____

How do shrimp hide from their enemies? (21)_____

What appendages do they use to bury themselves? (22)_____

What appendages protrude above the sand when the shrimp are buried? (23)_____

Why are having stalked eyes important to the shrimp? (24)_____

Can the eyes move or are they fixed in one position? (25)_____

How might this be an advantage to the shrimp? (26)_____

What other behavior patterns have you observed? (27)_____

When shrimpers fish for shrimp, would it be better for them to have a net that drags along the bottom or at the top of the water? (28)_____

CONCLUSIONS

List three anatomical adaptations a shrimp has for protection.

(29)_____
(30)_____
(31)_____

List three behavioral adaptations a shrimp has for protection.

(32)_____
(33)_____
(34)_____

Now that you know a great deal about shrimp, see if you can get inside its head. In a short paragraph, describe the world from a shrimp's point of view. Try and think as shrimpy as possible and tell us how you might feel if you were a shrimp.

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MEASURING WITH A MICROSCOPE

INTRODUCTION

From the standpoint of communication, it is essential that the student of marine science be able to view a microscopic organism and record its size. With this datum, the relative size of the organism in a drawing or photograph can be determined.

COMMENTS TO THE TEACHER

There are special attachments and devices for microscopes providing greater accuracy in measurement. Some fit beneath the ocular lens and are provided with grids and ruled lines. Many microscopes have a built-in micrometer in order to measure depth or thickness.

This particular lab procedure is given with full realization of its limitations in accuracy. It is believed, however, that the student can gain a better understanding of not only size relationships, but also he will have at his disposal a method of measurement regardless of which type microscope he uses. Not all labs provide ocular measuring discs. If one type microscope is used throughout, the teacher may wish to compile all estimations submitted by the students and decide upon a standard figure for both the low and high power sizes.

PROBLEM UNDER INVESTIGATION

A. To determine the size of a microscopic organism.

B. To indicate the relative size drawing or photograph of a microscopic organism with an accompanying scale.

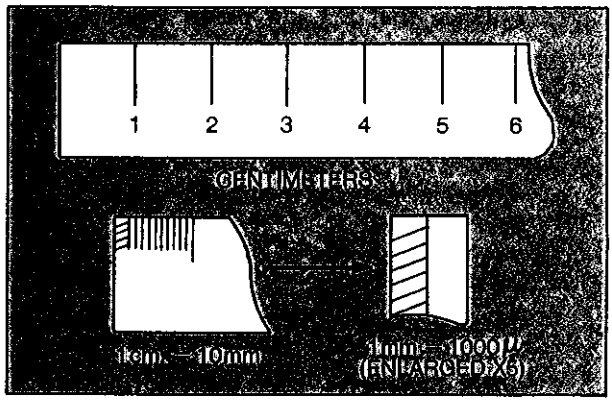
TO THE STUDENT

Examination of any marine organism is incomplete without reference to the size of the organism. It becomes the responsibility of a student of marine science, to learn the metric system and be able to measure the length of a copepod as well as the length of a shark.

The metric system enables the biologist to measure extremely small organisms in units of less than 1 millimeter in size. For purposes of this course, organism sizes under the microscope are expressed in microns (μ). 1 micron = .001 mm. In other words, there are 1000 microns in one millimeter.

Inspect your microscope and record:

1. Ocular magnification_____
2. Low power objective magnification _____
3. High power objective magnification _____



Microscopes differ as to ocular lenses available and number of objective lenses on the turret. So that we have a starting point for this exercise, we will assume that the ocular is 10x, the lower power is 10x and the high power is 44x. If your microscope differs from this, you may adapt by substituting your magnifications. The method is the same, regardless.

MATERIALS

Compound microscope
A plastic ruler

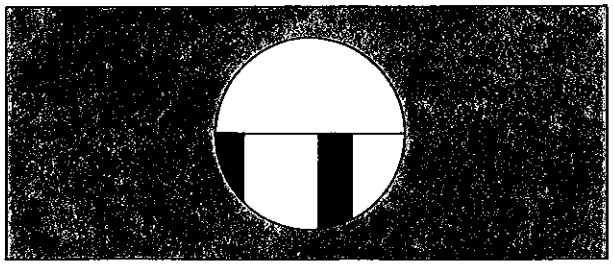
PROCEDURE

PART I: How to measure a microscopic organism

A. To determine the size of the microscopic field.

1. Measuring the low power field of vision.
   a. Place the plastic ruler on the stage of the microscope so that the metric edge covers one-half of the light-opening hole.
   b. With the low power objective in place, focus on the edge of the ruler.
   c. The distance between two lines (you will probably see no more than 2 lines) represents one millimeter.

d. Move the ruler so that the middle of one line is at the edge of the field of vision:



e. You will see that about one and one-half spaces will fit across the lower power field of vision.

f. Since 1 mm equals 1000 microns, we can estimate that the low power field of vision equals about 1500 microns (1500 μ) Note: These dimensions may vary with different microscopes.

2. Computing the high power field of vision.

a. To begin, we have the following data:
Low Power:
Ocular = 10x
Objective = 10x
Magnification = 100x (10x ocular times 10x objective)
Field diameter — 1500 microns (μ)

b. By inspection, we find that the high power objective is 44x. Therefore, the magnification under high power is 440x (10x ocular times 44x objective).

c. We also know that when we switch to high power, two things occur:
i. the size of the field is decreased.
ii. the amount of light coming through is insufficient and must be increased.

d. In fact, the high power viewing arch in this case will be 4.4 times smaller than the low power. A ratio is obtained be comparing magnifications. This is an inverse ratio, because as the magnification is increased, the size of the field decreases.

e. We now have the following data:
High Power:
Ocular = 10x
Objective = 44x
Magnification = 440x
Field diameter =341 microns (μ)

f. Computation

$$\frac{\text{100 (low power magnification)}}{\text{440 (high power magnification)}} = \frac{1}{4.4}$$

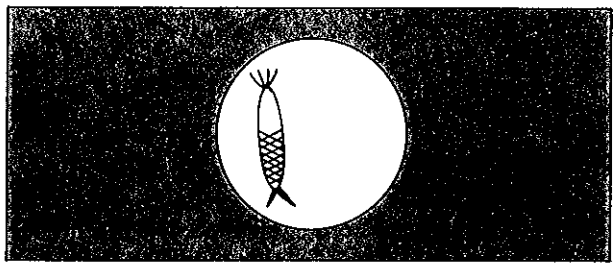
then $$\frac{1}{4.4} = \frac{\text{Diameter of High Power Field}}{\text{1500 (Diameter of Low Power Field)}}$$

thus, solving the equation for the diameter of a high powered field, you arrive at: 340.9

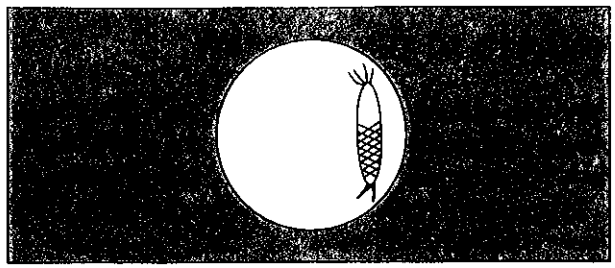
B. To determine the size of a microscopic organism.

1. Estimating the size of an organism under low power.

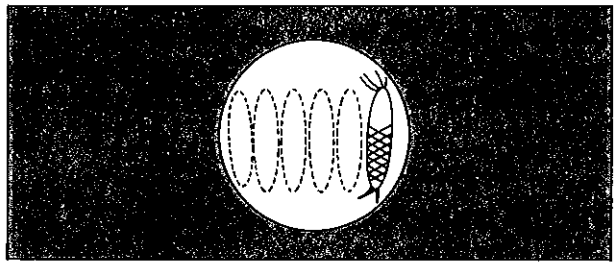
a. Suppose you are viewing an organism under low power and wish to estimate its width, for instance.



b. Move the slide so that the organism is oriented thus:



c. Now, by inspection, how many of these organism could fit across the field:



d. If, for example, 7 of these could fit across the field, then the width of each organism would be:

$$\frac{1500}{7} = 214.2$$

2. Estimating the size of an organism under high power.

a. Refer to parts A1 and A2. Naturally, the low power field size must be determined before proceeding here.

b. In "Part I. A. 2. f." we found the diameter of the high power field to be 341 μ.

c. Again, as done in "Part I. B. 1. c." we must estimate how many organisms could fit across the diameter of the high power field of vision.

d. This done, we divide the diameter (341 μ) by the number of organisms that will fit across the field.
e. The answer you get will be the actual size of the organism . . . in microns.

PART II: How to indicate the size of an organism on a drawing. Note: Whether the organism is microscopic or macroscopic (large enough to observe with the naked eye), the procedure is fundamentally the same.

Step I

A. Microscopic organism
Compute the actual size of the organism from instruction in Part I.
B. Macroscopic organism
Measure with a ruler, meter stick, calipers, etc.

Step II

Draw or photograph the organism.

Step III

A. Microscopic organism
Measure the drawing, in millimeters and convert to microns (1mm — 1000 μ)
B. Macroscopic organism
Measure the drawing in millimeters.

Step IV

Making sure that the acutal measurement and the drawing measurement are in the same unit of measurement, i.e. both millimeters or both microns.
Do this:
Divide the size you got in your drawing by the size you got for the actual organism.

Step V

The answer you get will be the size relationship on "scale" between your drawing and the actual size of the animal, the cell, or "whatever."
If your answer comes out to be 1 or more, it would be expressed:
x1,x1½x6x10,x250,etc.
If your answer is less than one, it is expressed as a fraction:
x½,x¼,x1/6,x1/5,etc.

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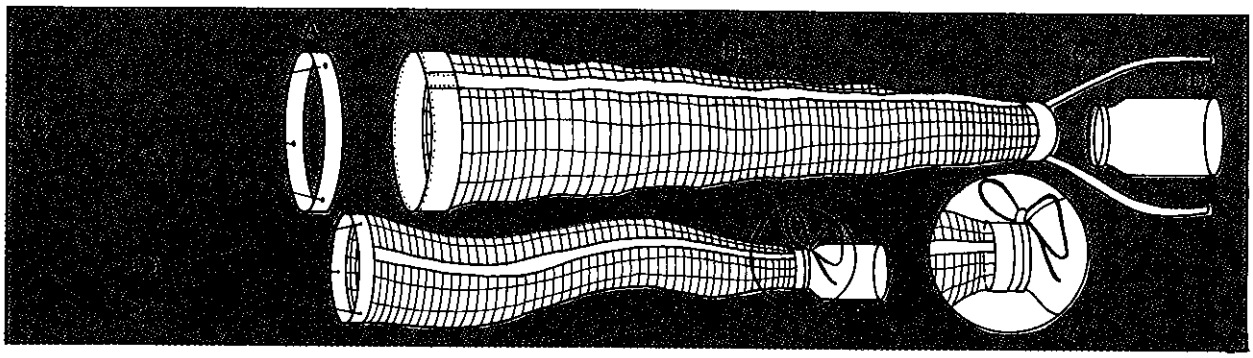
PLANKTON

TO THE TEACHER

Since a plankton net is essential for this study, instructions are included for construction of a simple plankton net.

Pulling the net behind a slowly moving boat (1-2 knots) is one standard method of collecting plankton. For surface sampling the net should tow just below the surface. For deeper sampling tie a weight to a separate line and fasten to the front swivel. Even throwing the net from a bridge or pier into a fast tidal current often produces excellent results.



A. Ring: Solidly constructed of brass, steel, etc. with diameter varying from 20 centimeters to 1 meter. Steel wire, chain or rope leaders are attached to ring and to a single swivel at the forward end.

B. Net: Long nylon stockings or silk bolting cloth with about 125-200 meshes to the inch are recommended. Reinforced with canvas around the openings.

C. Ties: Sew on two canvas ribbons along the length of the net as shown. The free ends serve to secure the collecting bottle.

D. Bottle: Sturdy plastic or glass bottle with deep threads at the neck. Size depends upon net diameter. Plastic mustard and ketchup dispensers, glass cherry, olive or babyfood jars, etc., are adequate.

Biological supply houses generally stock an assortment of plankton nets and accessories. Adapters are available to be permanently fastened into the net opening so that collecting bottles can be screwed into place rather than tied. But, best results will be achieved with the smallest mesh size, whether the net is purchased or constructed. A more stimulating approach may be to let each student team construct their own expendable gear for the exercise and see how each does.

This particular activity centers around the collection and use of a concentrated sample which can then be diluted and distributed to an entire class. If time is available the following types of comparative samples can easily be substituted:

1. Individual samples from the same area.
2. Samples from different areas.
3. Samples taken at different times during the day (with reference to light and tide).
4. Samples taken at different depths (vertical sampling).

The sample needed for this laboratory should be taken in the early morning before school . . . to be used the same day, or it may be collected the night before and kept in a cooler with ice. This plan will almost insure students the rare opportunity to observe living plankton. Place sample in a larger jar or bucket and aerate gently. Dilute with a known quantity of filtered sea water. When ready to begin the lab, divide the sample into equal portions. Make available the following information to students:

Example:

a. date and general location of sampling — 1/21/80 N. Tampa Bay
b. time of day — 0600 hrs.
c. wave activity and temperature (optional) — light chop, 24°C.
d. tide — low (incoming - outgoing)
e. depth of sample — surface
f. tow time (15 minutes is adequate for a good sample) — 15 minutes.
g. mesh size — 125/inch
h. diameter of net opening (in meters) — .2
i. estimated distance net towed (in meters) — 1500
j. number of ml of team sample — 1
k. dilution 2:1

Three periods are needed to complete all phases of the exercise. Each day includes a separate purpose, set of procedures and observation guides.

First Day observation of living plankton: student should

become aware of (a) dominant forms, (b) methods of locomotion (c) variation in shapes.

Second day: Measuring and classifying plankton (a) obtaining representative measurements of dominant forms (b) identifying common types.

Third Day: Total Sample Analysis (a) learning how to count plankton (b) estimating size of population in comparison to total volume of water sampled.

TO THE STUDENT

There exists in the oceans of the world, in seas, bays, lakes and in nearly every other natural water body, a population of organisms so immense that it defies counting! Everyone, while swimming has probably brushed up against millions of these creatures without being aware of their presence. No doubt some even swallowed a large number while learning to keep mouths closed underwater!

Although limited studies of this population were made before 1887, it was not until that year when an oceanographer, Victor Hensen, first proposed a name for this vast assemblage . . . plankton. The term refers to those plants and animals mostly microscopic in size, that are made "to wander or drift" (Hardy, 1961), under the influence of ocean currents and tides. Even though many planktonic forms have the ability to swim, their efforts in the presence of oceanic water movements are generally too feeble and in vain. Animal members are named zooplankton. With the exception of marine mammals and reptiles, nearly every creature in the sea spends either a part or all of its life drifting about. Eggs, larvae, and juveniles of most invertebrates and fishes and even some adult forms are common. Copepods (crustaceans) are the most abundant and universally distributed animals in the plankton.

Phytoplankton (plants) are more numerous than their animal counterparts, and are best represented by the microscopic diatoms that form the vast bulk of the ocean's vegetation.

Because of their numbers, wide distribution and beneficial biological activities, plankton are considered the most important inhabitants of the marine world with all forms of life directly or indirectly dependent upon them. Plankton are basic to the food chains of all marine life, Sponges, tube worms, clams and sea squirts filter out sea water to gather them. Herring fisheries use plankton indicators to predict the potential catch (Russell and Younge, 1963). Giant baleen whales, over 100 feet in length and reaching fantastic weights of 150 tons, feed exclusively on plankton. Diatoms are an important source of vital oxygen and proteins which animal life cannot snythesize but require. Without plankton the seas would surely be a wet desert!

OVERALL PURPOSE

The purpose is to observe and analyze a plankton sample. This will be done in three different exercises.

MATERIALS

Compound microscope
Dissecting scope
Petri dish
Preservative (3.5% formalin)
Centimeter grid or graph paper
Sample bottle and label
Plastic metric ruler
Eye dropper
Standard slides and coverslips
Concavity slides
Diluted plankton sample for each team

EXERCISE I

PURPOSE

To observe living plankton.

PROCEDURE

1. The teacher will provide the recorded conditions under which the plankton sample was made. Enter this information on the data sheet provided.
2. Obtain diluted portion of the plankton sample and pour into a petri dish, gently swirl the contents to distribute.
3. Observe with a dissecting microscope. Scan the entire field.
4. Many organisms are too small to be seen with the dissecting scope. Prepare wet mounts of the sample and observe under a compound microscope with low and high power objectives. Do not discard any part of the sample. Empty wet mounts back into petri dish. Rinse with medicine dropper of water.
5. Do not record any observations today. Look for:
   a. most abundant organisms
   b. variations in shape, color and swimming abilities.
   c. types of appendages
   d. chlorophyll-containing organisms
   e. eggs
   f. larval and juvenile forms of crustaceans and fish (see pictoral guide to the plankton)
6. Preserve sample in 3-5% formalin before leaving. Label sample bottle.

EXERCISE II

PURPOSE

To draw, measure and record characteristics of dominant organisms in plankton sample.

PROCEDURE

1. Select the most common organism from your preserved sample. Prepare a wet mount and view with low power (or high power).

PLANKTON LABORATORY

Data Sheet

I. Record the following conditions pertaining to the sample under study:

a. date _____ d. tide _____ g. mesh size _____

b. location _____ e. depth _____ h. diam. net opening _____ meters

c. time of day _____ f. tow time _____ i. distance net towed _____ meters

j. dilution of your sample _____ k. volume of your sample _____ ml

II.

organism #	1	2	3	4	5	6
drawing						
actual size						
drawing size						
magnification						
identification outstanding features ±? coloration? common and/or scientific name						

III. a. volume of water which passed through the plankton net _____ M3

b. number of macroscopic organisms in your sample (T) _____

c. number of macroscopic organisms in original sample (Q) _____ M3

2. Record the following information on data sheet:
   a. a detailed penciled drawing of the specimen:
   b. measured actual size, in microns
   c. measured drawing size, in microns
   d. magnification of drawing
   e. identification

3. Repeat this procedure with as many different specimens as time permits. Record all observations.

4. Do not discard any portion of your sample. Empty wet mounts back into sample bottle. Rinse with dropper of water.

EXERCISE III

PURPOSE

To compute the size of the plankton population in comparison to total volume of water strained.

1. Compute total volume of water strained as follows;

A plankton net with a measured net opening was towed behind a boat for a measured distance. How much water passed through this net? The total number of organisms collected from this tow represents the concentration of plankton in that volume of water.

a. To compute water volume which passed through net towed behind a boat:

$$M^3 = \frac{\pi D^2}{4} \times L$$

M^3 = volume of water in cubic meters
D = diameter of net in meters
L = length of tow in meters

for example: a net with a diameter of 2 decimeters was towed 1500 meters:

$$= \frac{3.14 \times 2^2 \times 1500}{4}$$

= 47.1 cubic meters

b. To compute volume of water which passed through net tossed from a bridge:

First determine length of tow:

$$L = \frac{Ti}{t} \times W$$

L = length of tow in meters
Ti = length of time net was immersed in water
t = time floating object took to pass width of bridge
W = width of bridge in meters

for example: a net strained water for 18 minutes. It took a cork .25 minutes (15 seconds) to pass under a bridge 12 meters wide. Thus:

$$L = \frac{18 \times 12}{.25}$$

L = 864 meters

. . . now compute water volume (assuming that net diameter is still 2 meters).

$$M^3 = \frac{\pi D^2}{4} \times L$$

$$= \frac{3.14 \cdot .2^2 \times 864}{4}$$

= 27.1 cubic meters

2. Compute the density of the original concentrated sample (number of macroscopic specimens/ _____ml) as follows;

The original sample contained _____ ml of liquid. It was diluted with an equal volume of sea water. You then received _____ ml of this diluted sample. How many macroscopic (visible) organisms are contained in: (a) your sample and; (b) the original sample. Diatoms and other microscopic forms have been eliminated from this count.

a. Pour your sample into a petri dish.
b. Determine the area of the dish.

$$A\ (cm^2) = \frac{\pi D^2}{4}$$

A = number of sq. centimeters covered by dish.

c. Place petri dish over a centimeter grid or graph paper with centimeters squares clearly marked. Distribute sample evenly over bottom of dish.
d. Select at random, five (5) squares and count all macroscopic organisms in each. To determine average divide total number of organims by 5.
e. To estimate total number of organisms in the petri dish, (thus in your sample):

$$T = \frac{t \times A}{g}$$

T = total macroscopic organisms in your sample
t = total count from random grids
g = number of grids counted
A = area, in cm^2 of petri dish

for example: 100 organisms were counted from 5 random grids. The area of the petri dish covered 78.5 cm^2.

$$\text{thus} = \frac{100 \times 78.5}{5}$$

= 1570 macroscopic organisms/ml diluted sample:

f. Compute total of macroscopic organisms in original concentrated sample:

$$Q = T \times \frac{V}{v}$$

where . . .
Q = total macroscopic organims passing through plankton net.
T = total number of organisms in your sample.
V = total volume of original concentrated sample.
v = volume of your sample.

for example: Ten (10ml) of sample was strained from 30 cubic meters (M^3) of sea water. You received 1 ml of

this and proceeded to count 1570 macroscopic organisms.

Thus:

$$\frac{1570 \times 10}{1} = 15{,}700 \text{ macroscopic organisms / } 30 \text{ M}^3$$

g. Compute the total number macroscopic organisms per cubic meter.

4. Record all information on data sheet.

QUESTIONS FOR CONSIDERATION

1. How are organisms which are not part of the plankton population classified?
2. What is the DSL?
3. Which environmental factors may influence the vertical migrations of plankton?
4. How do minute and delicate plankton withstand the crushing pressures of deeper waters?
5. How do diatoms and copepods figure into the food chains of marine organisms?
6. How do baleen whales feed on plankton?
7. Are there ocean areas in the world devoid of plankton?
8. What was your impression of the abundance of diatoms in the plankton sample? Copepods?
9. Which phyla of organisms were best represented in your sample?
10. What happens to plankton which "drift" into waters where conditions of salinity, oxygen, or temperature are unfavorable? Explain answer.
11. What are some limitations to using plankton net in sampling populations of organisms?
12. What are some limitations to the methods used in counting the density of plankton populations? What are other methods of estimating the density of plankton?
13. What is meant by "standing crop of plankton"?
14. What do the prefixes, "holo-, mero-, and mano-" refer to in reference to plankton? Give examples.
15. What adaptations do plankton have for moving?
16. Describe biological magnification of a pesticide such as DDT, which is now found in every part of the world ocean and is taken up by plankton organisms. Assume a concentration of one part per ten billion (1 x 10 -10) in sea water.

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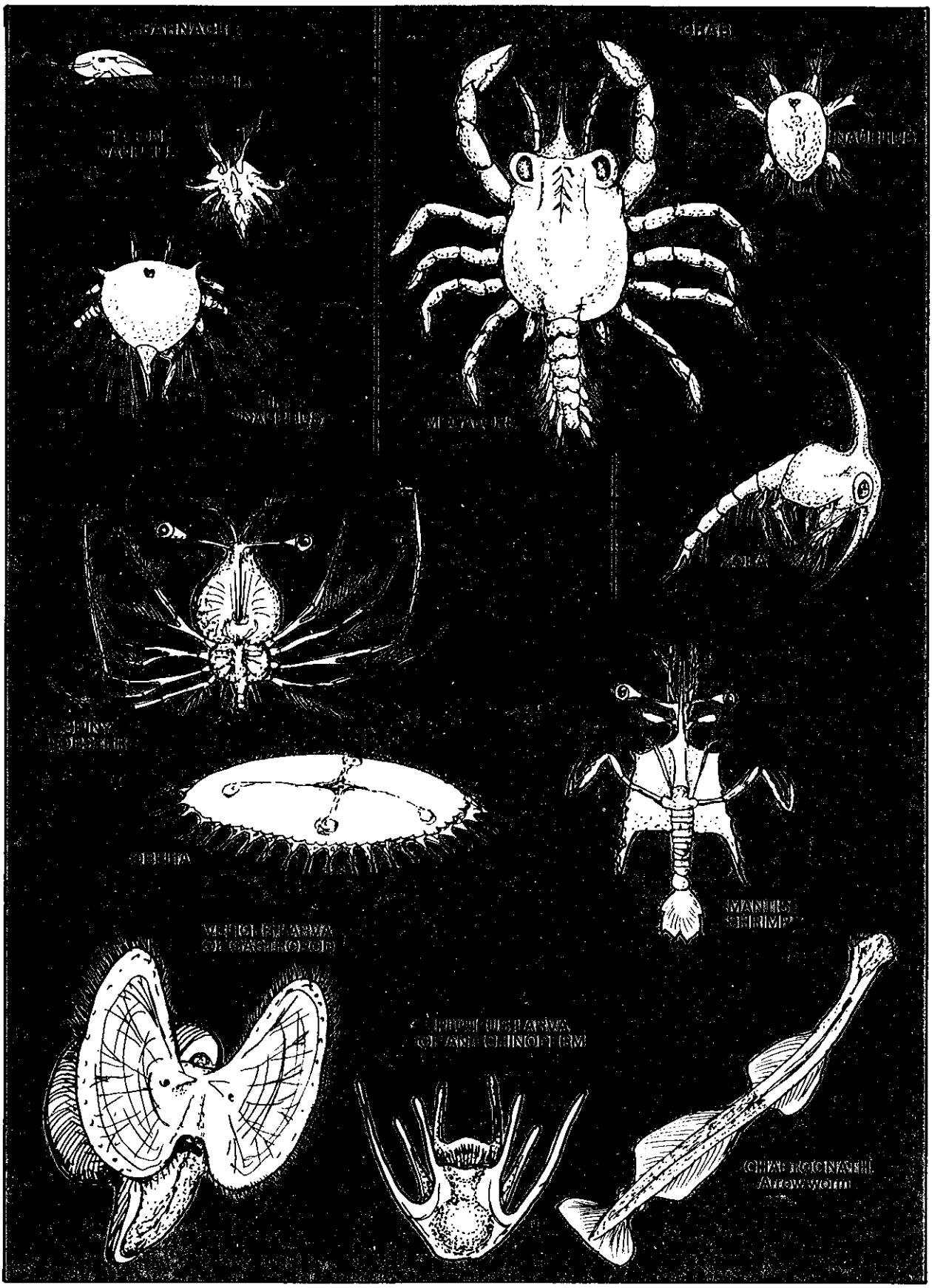
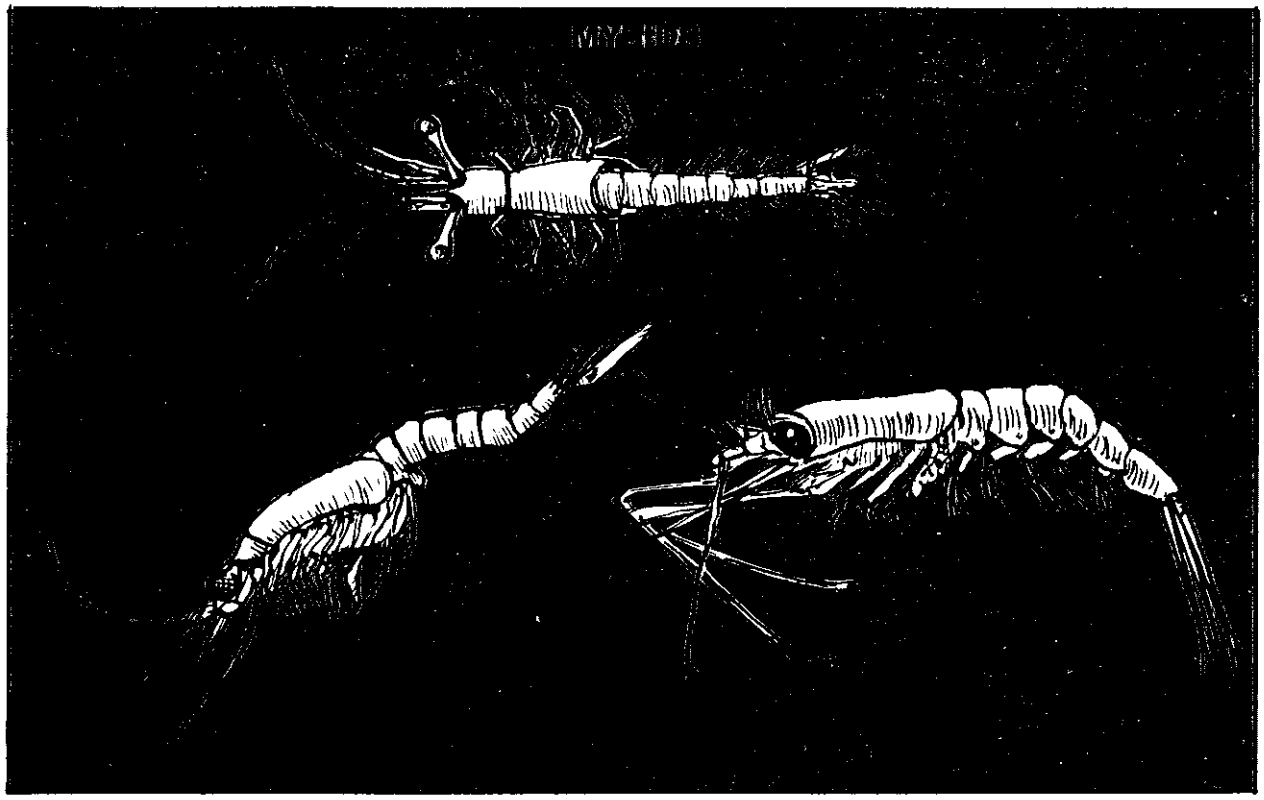
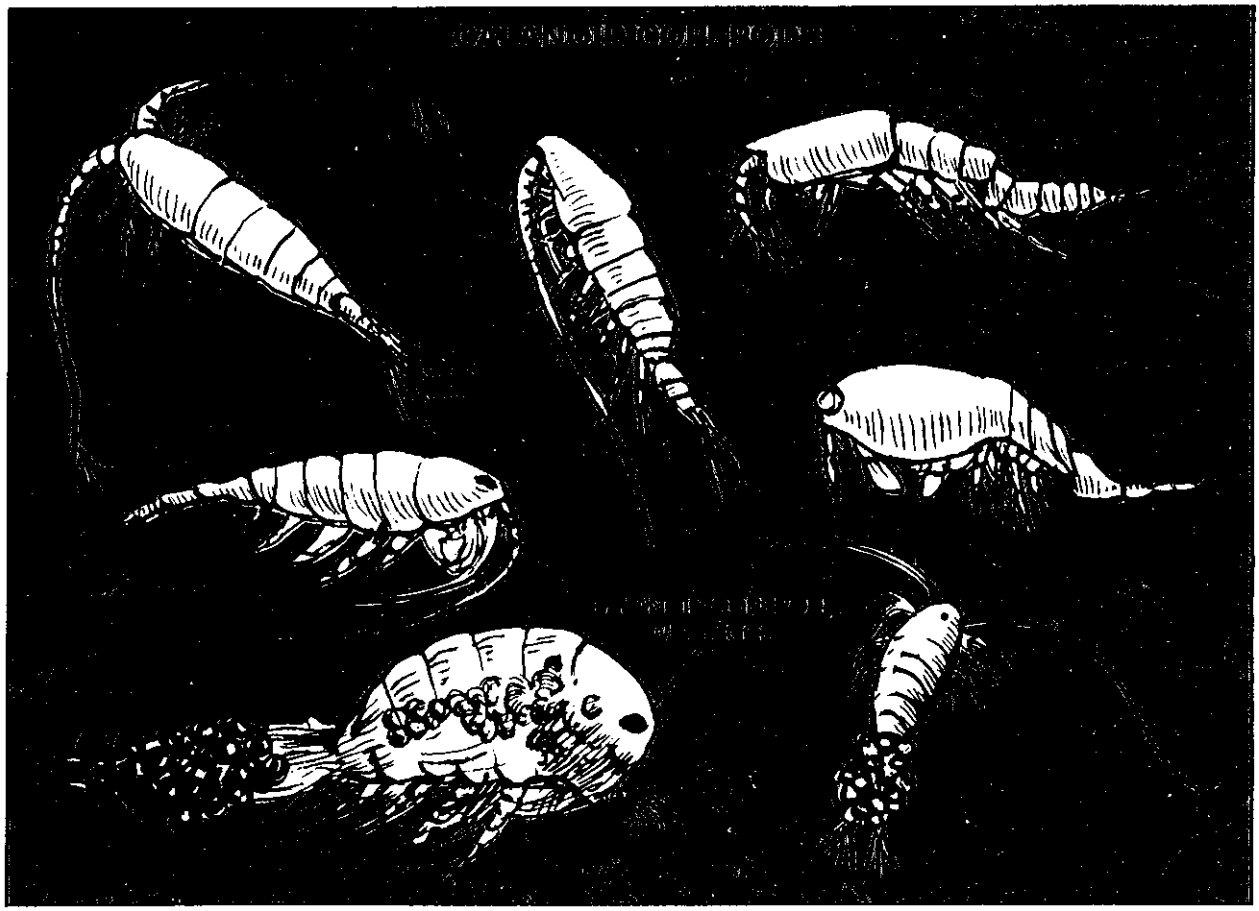
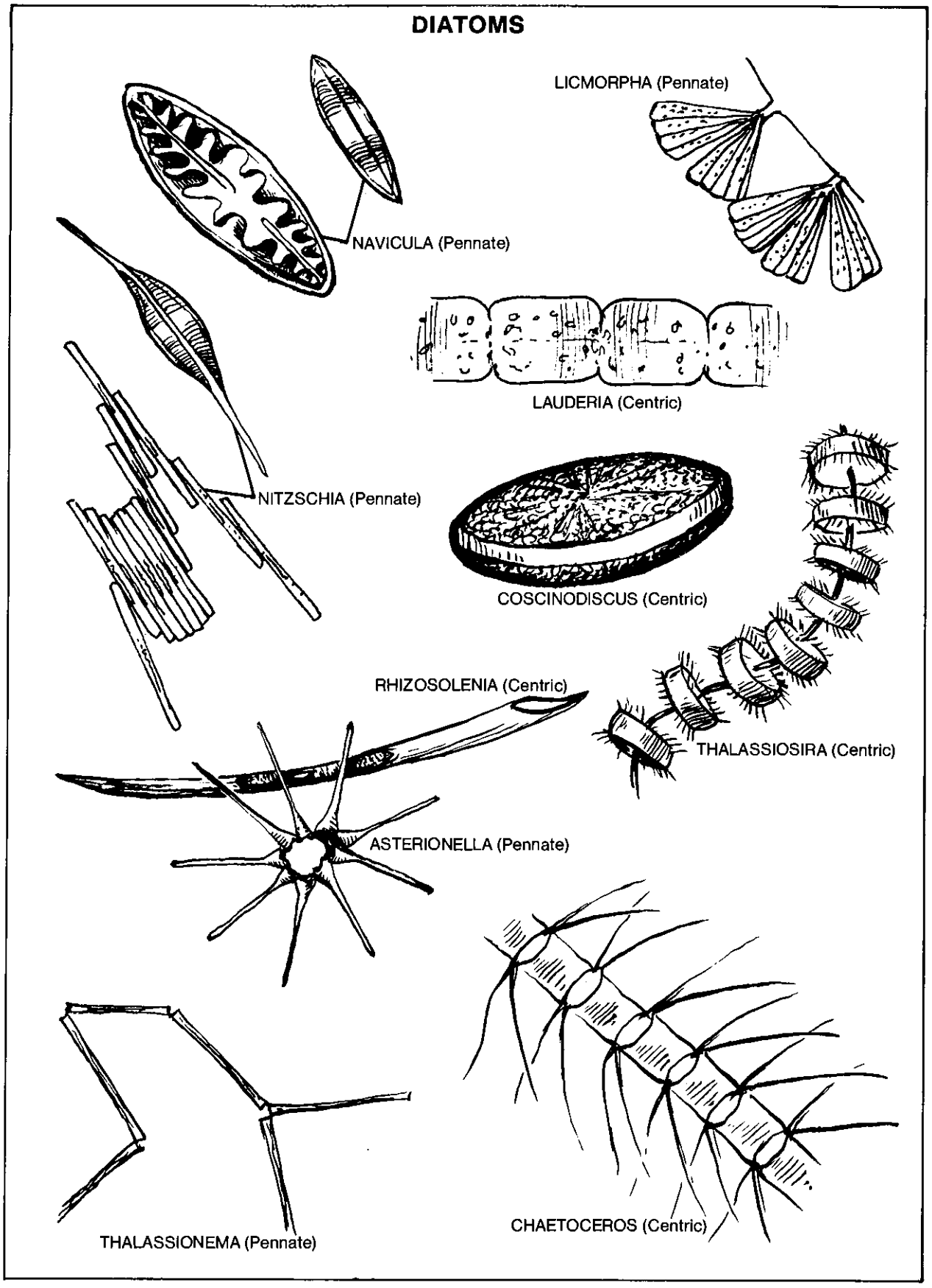
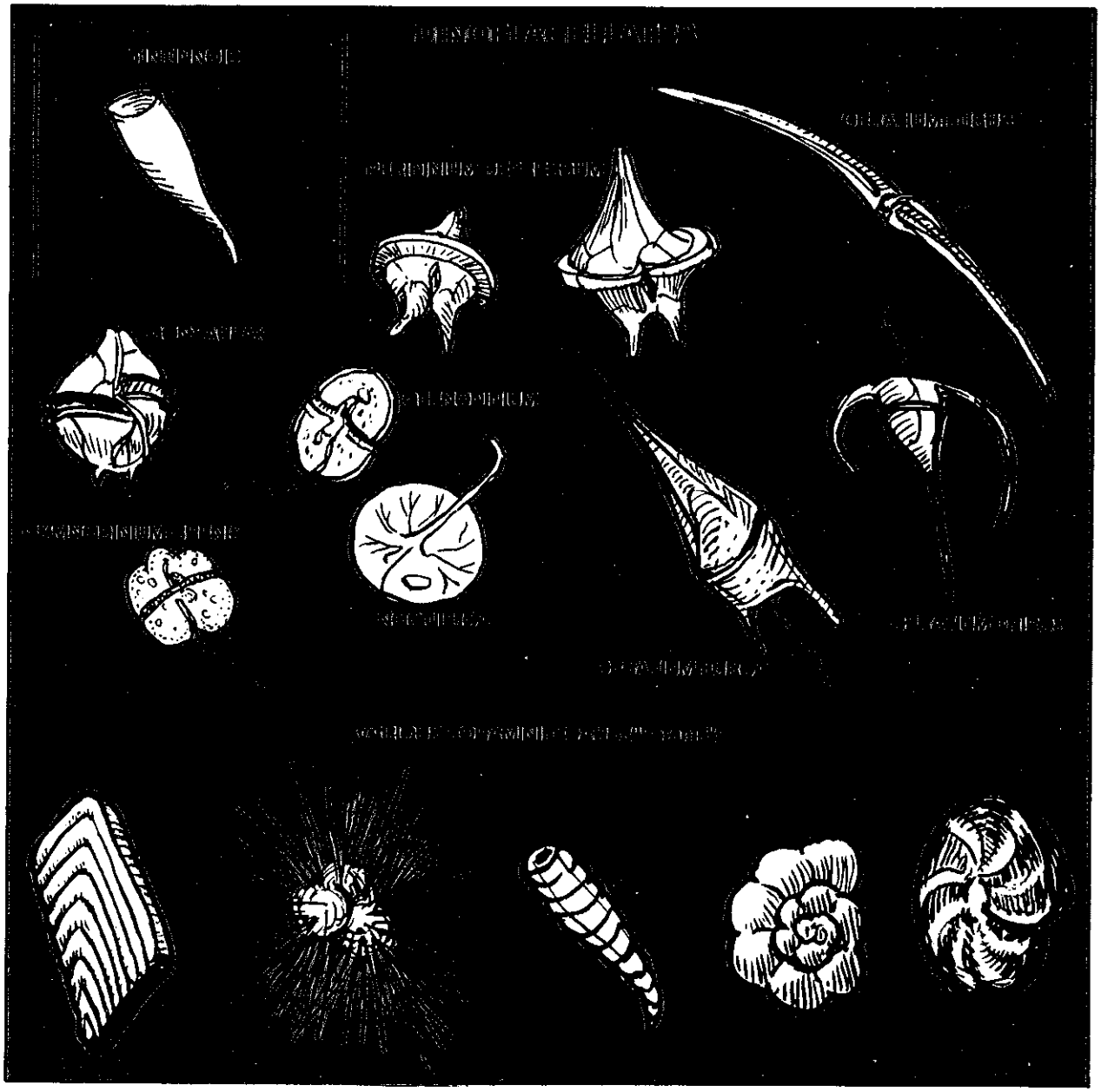
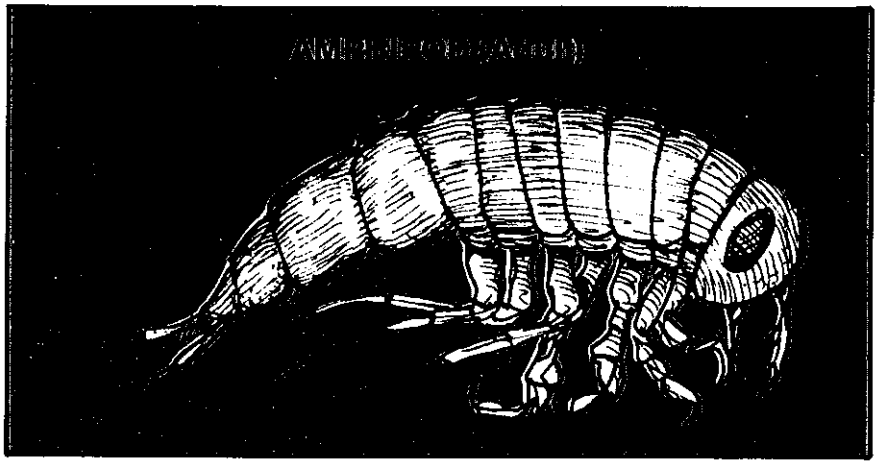

CHAETOGNATH
Arrow worm






DIATOMS
LICMORPHA (Pennate)
NAVICULA (Pennate)
LAUDERIA (Centric)
NITZSCHIA (Pennate)
COSCINODISCUS (Centric)
RHIZOSOLENIA (Centric)
THALASSIOSIRA (Centric)
ASTERIONELLA (Pennate)
THALASSIONEMA (Pennate)
CHAETOCEROS (Centric)





LIVING WORLD WITHIN A SPONGE

TO THE TEACHER

This study will help students realize one important role of sponges in the marine environment . . . that of providing shelter for a horde of creatures.

Fresh sponges are recommended for this activity. Specimens may be collected one day, enclosed in strong plastic bags, stored in a refrigerator overnight and used the next day. Further delay will prove highly undersirable . . . sponges decompose rapidly!

In order not to destroy the local population of sponges or if sponges are not available in your area, you can make your own sponge habitat by filling a net dive bag with an assortment of sponges purchased in a hardware store. The bag can be anchored to the bottom in an area of high productivity. The sponges should remain for approximately 10 or more days allowing the habitat to develop. Some sponges may do better than others and you may want to try different locations for different time periods.

Small sponges could be assigned to one student, while larger specimens would prove enough material for a team or even an entire class.

TO THE STUDENT

Sponges are strange creatures and not at all like the typical animals that are familiar. They have no head, body, arms, legs or any other obvious structures that are generally associated with animals. All that is apparent are the countless pores which riddle their strong-smelling tissues.

Sponges are microscopic during their larval stage and drift about as part of the plankton population. Their wanderings are short-lived as they soon settle to the bottom and become anchored . . . destined to spend the rest of their days in one place!

Although no animal looks to the lowly sponge as a source of food (try tasting a sponge sometime), many creatures take up residence in their intricate network of canals. Rachel Carson (1955) wrote: "One such permanent lodger is a small shrimp — one of the group known as snapping shrimp because of the sound made by snapping the large claw. Although the adults are imprisoned, the young shrimp, hatched from eggs adhering to the appendages of their mothers, pass out with the water currents into the sea and live for a time in the currents and tides, drifting, swimming, perhaps carried far afield. By mischance they may occasionally find their way into deep water where no sponges grow. But many of the young shrimp will in time find and approach the bulk of some loggerhead sponge and, entering it, will take up the strange life of their parents. Wandering through its dark halls, they scrape food from the walls of the sponge. As they creep along these cylindrical passageways, they carry their antennae and their large claws extended before them, as though to sense the approach of a larger and possibly dangerous creature, for the sponge has many lodgers of many species other shrimps, amphipods, worms, isopods and their numbers may reach into the thousands if the sponge is large."

PURPOSE

To discover which types of organisms are residents of sponges.

MATERIALS LIST

Dissecting kits
Sorting jars
Dissecting microscopes
Balances and rulers

PROCEDURES

1. Carefully slice off a thin section of sponge tissue. Examine for animal life.
2. Sort out creatures according to phyla, class, etc.
3. Repeat this procedure until entire sponge has been dissected and all inhabitants removed and sorted.

SUGGESTED MATHEMATICAL COMPUTATIONS

1. Record total population found in sponge.
2. Compute per cent of total population for each species by numbers and weights.
3. Compute weight-length ratio for each taxonomic group.

SUGGESTED GRAPHIC REPRESENTATION OF DATA

Prepare a graph showing the following:
1. Frequency distribution of sponge population.
2. Weight distribution according to species.

QUESTIONS FOR CONSIDERATION

1. How could one account for the presence of animal life within a living sponge? How would you classify this relationship?
2. What kinds of competition would go on between the tenants?
3. Is the sponge harmed in any way because of these tenants?
4. Can you attach any economic importance to the sponge's role of providing shelter for a multitude of creatures?
5. Do you think that the population inhabiting a sponge changes with seasonal conditions? Why?

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HORSESHOE CRAB

TO THE TEACHER

This is a laboratory exercise requiring 1 to 3 periods depending upon the following: 1 period if only preserved specimens are available; 2 to 3 periods if both living and preserved specimens are available.

The horseshoe crab is a hardy animal. It can be collected several weeks ahead of the time planned for study. Proper care and feeding should be used to insure a healthy specimen. After completion of the lab, return all living animals to the environment.

The horseshoe crab is found in very shallow water of bays and estuaries, sometimes by the thousands in the spring and early summer.

TO THE STUDENT

The horseshoe "crab" is in the phylum Arthropoda but not in the class Crustacea as are many common occurring crabs (see Taxonomy of Marine Organisms). The horseshoe crab is in the class Merostomata which are believed to be ancestors to the Arachnida which contain spiders. The horseshoe crab is referred to as a "living fossil" since it has remained virtually unchanged over the past 400 million years.

The horseshoe crab (king crab) feeds on worms, mollusks, bottom-dwelling algae, plus other material it scavenges from sandy or muddy beaches and bottoms with its chelicera (front feeding pincers.)

At one time the horseshoe crab was considered a pest along the upper east coast of the United States and a bounty of one cent was placed on each animal collected. The animals were then dumped on the town trash pile.

In other areas, they have been collected in great quantities and ground for fertilizer or chicken feed supplement.

PURPOSE

To gain a knowledge of the external anatomy and behavior of an interesting arthropod, *Limulus polyphemus,* the horseshoe crab.

MATERIAL

Preserved specimens
- Dissecting pan or tray
- Probe
- Horseshoe crab

Living specimens
- Aquarium
- Light source, and small pieces of shrimp

PROCEDURE

Place the living horseshoe crab in an aquarium with a sand or gravel bottom for the following observations:

a. Coloration of exoskeleton
b. Method of locomotion
c. Method of righting itself if turned upside down.
d. Attraction to light: The light should be oriented from a corner position to determine if there is a response.
e. Method of burrowing
f. Number and kind of attached organisms
g. Feeding habits
h. Determination of sex
i. Method of swimming

If preserved specimens are at hand, the student should locate and record a physical description of the external characters shown in figures 1 and 2.

DATA RECORDING

Record all observations on the living specimen and repeat observations as many times as possible during one laboratory period, while checking for possible variations in behavior patterns.

FORMULA AND MATHEMATICAL COMPUTATIONS

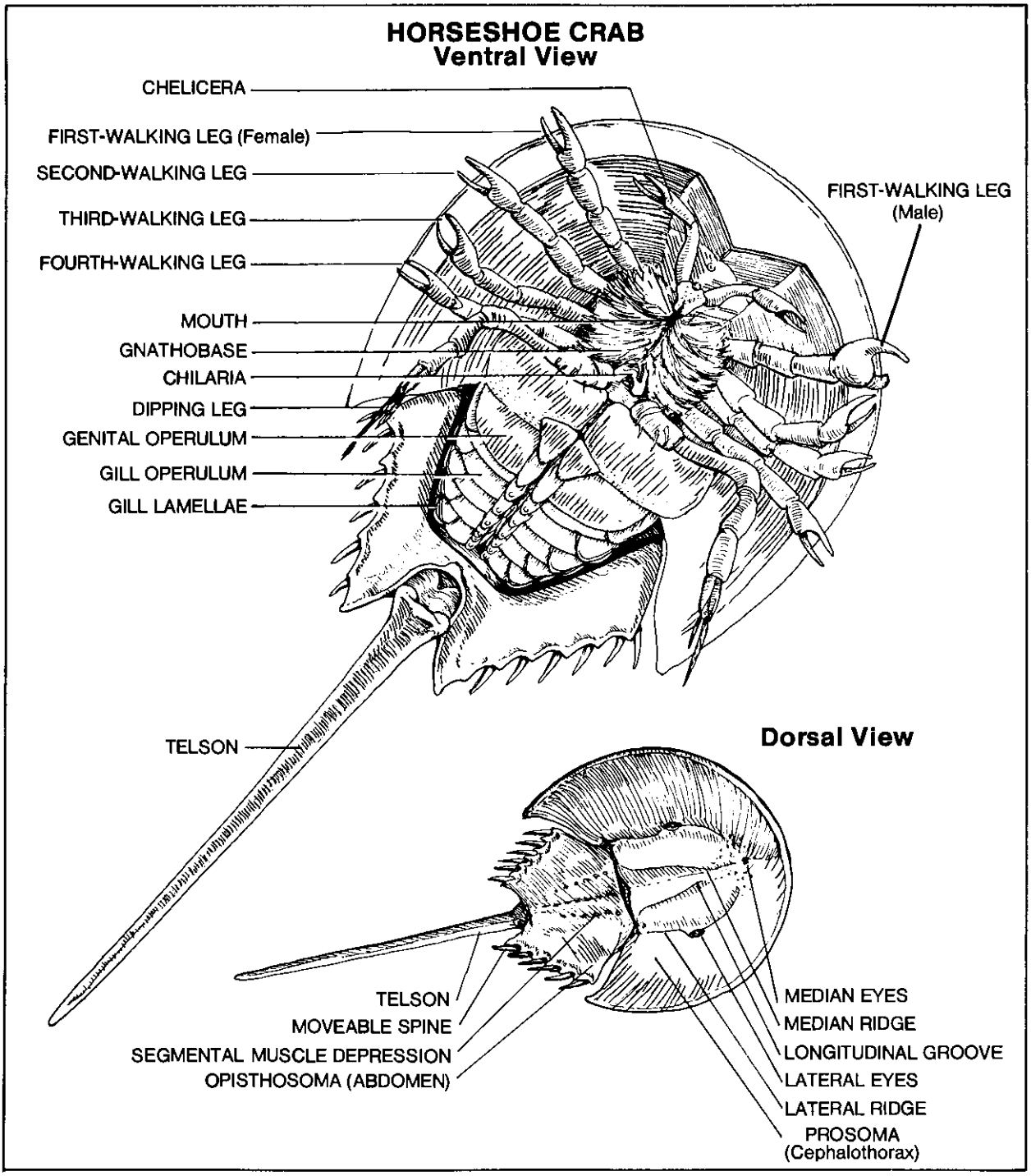
The male genital openings are round and the female genital opening is a transverse slit. The number of males and females used by the class should be computed as percentages.

$$\frac{\text{Number of males (females)}}{\text{Total number of specimens}} = \text{\_\_\_\_\_}\%$$

GRAPHIC ANALYSIS AND CONCLUSIONS

It may be feasible in your area (providing horseshoe crabs are plentiful) to carry out one of the following suggested experiments.

1. Survey the living population to determine how many


HORSESHOE CRAB
Ventral View
CHELICERA
FIRST-WALKING LEG (Female)
SECOND-WALKING LEG
THIRD-WALKING LEG
FOURTH-WALKING LEG
MOUTH
GNATHOBASE
CHILARIA
DIPPING LEG
GENITAL OPERULUM
GILL OPERULUM
GILL LAMELLAE
FIRST-WALKING LEG (Male)
TELSON
Dorsal View
TELSON
MOVEABLE SPINE
SEGMENTAL MUSCLE DEPRESSION
OPISTHOSOMA (ABDOMEN)
MEDIAN EYES
MEDIAN RIDGE
LONGITUDINAL GROOVE
LATERAL EYES
LATERAL RIDGE
PROSOMA
(Cephalothorax)

specimens have a growth of algae or barnacles present and prepare a graph of size (carapace diameter) versus number of growths present.

2. Try to find a population of small, white worms living on the horseshoe crab. These flatworms, named *Bdelloura* (del-oura), are not parasites and have a mutualistic relationship with the horseshoe crab.

3. Map the locations on the body at which *Bdelloura* (a marine flatworm) occurs and compute the frequency with which it occurs at each point.

4. Measure carapace diamater and telson length to be used in preparing a graph.

QUESTIONS FOR CONSIDERATION

1. Why and how does the horseshoe crab molt?

2. What function could the movable spines located on the opisthosoma (abdomen) have?

3. What potential economic importance could this creature have?

4. What are the ancestral origins of the horseshoe crab?

5. What are the functions of the gnathobase of each appendage?

6. Do the median ridges offer protection for the compound eyes?

7. Does the complete body armor for internal organs and appendages account for the fact that they have remained unchanged over some 400 million years?

8. What predators must the horseshoe crab face?

9. What provides competition for the horseshoe crab in its environment?

10. Why do barnacles and algae seldom attach to the carapace of the horseshoe crab?

11. Why does the horseshoe crab burrow in the sand?

12. Why is *Bdelloura* attached in the horseshoe crab? Of what benefit is the *Bdelloura* to the horseshoe crab? Of what benefit is the horseshoe crab to *Bdelloura?*

13. When in the horseshoe crabs life cycle is it most vulnerable to predation?

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STATISTICAL METHODS

INTRODUCTION

It is strongly suggested that this section, if applicable to the teaching situation, be presented as early in the semester as possible since many of the laboratory and field exercises call for some degree of statistical analysis. If copies are made for each student, less than one week's time is required to present this unit.

Being able to organize large amounts of data into meaningful terms is a difficult task for the average junior or senior high school student. Yet, an increasing number of laboratory and field activities stress, and frequently result in, an overwhelming pile of quantitative information. For this reason, a basic understanding of statistical methods should become an important part of the secondary science curriculum.

Even a superficial knowledge of statistics, as included in this section, will prove to be a definite asset to students who are in need of a tool to relieve the tedious burden of recording, classifying and interpreting data. It provides them with a step-by-step method of gleaning order and meaning from a mountain of numbers.

A fictitious case study is employed to illustrate how statistics are used in fishery biology and, at the same time, to teach students a sequential procedure to follow whenever a statistical treatment of information is needed.

CASE

Fishery biologists are continually searching for new methods to improve present yields of proteins from the sea to satisfy the demands of an increasing national and world population. One virtually untapped resource, as far as the United States is concerned, is the huge schools of squid reported off our continental shelves.

Japan leads the world's squid fisheries with more than 600,000 tons reported (Walford, 1958). During this same period (1952-53) North American fishermen reported landing about 6,500 tons. Lane (1960) stated, "... it is probably true to say that the annual catch of cephalopods throughout the world today is in the region of one million tons, or about one pound for every man, woman, and child on earth."

Lane continues, "... In fact the cephalopod population could probably stand several times the present amount of fishing."

Fishery biologists undertook a survey off the Florida east coast to determine the abundance and size distribution of the common squid in that area. The data presented below was the result of one sampling at a depth of 25 meters:

size (in inches)	number	size (in inches)	number
2	1	20	10
3	3	22	14
5	1	23	10
6	4	25	10
7	2	26	5
8	9	27	10
9	1	28	8
10	4	29	10
11	10	30	2
13	8	31	3
16	3	32	4
17	4	35	2
19	12		

At first glance the above data may appear to have limited value. However, as biologists proceed to transfer this information to statistical form its meaning and importance will begin to emerge.

In steps 1-9 in the procedure you will find the average size (mean) of the squid caught. Also you will find the size range of more than 2/3 of the sample. This will give an indication of the number and sizes of squid caught in future under these conditions. To the commercial fisherman, this data will give him an indication of what he can expect to catch.

PROCEDURE

Steps 1-9 refer to this statistical table . . . column by column.

A	B	C	D	E	F	G
i=5	X	f	fX	Δx	Δx^2	$f\Delta x^2$
31-35	33	9	297	+13	169	1521
26-30	28	35	980	+ 8	64	2240
21-25	23	34	782	+ 3	9	306
16-20	18	29	522	- 2	4	116
11-15	13	18	234	- 7	49	882
6-10	8	20	160	-12	144	2880
1- 5	3	5	15	-17	289	1445
		N=150	ΣfX=2990			$\Sigma f\Delta x^2$=9390

mean (M) = $\Sigma\frac{fX}{N} = \frac{2990}{150} =$ 19.9 or 20 inches

Standard Deviation (SD or σ) =

$$\sqrt{\Sigma \frac{f\Delta x^2}{N}} = \sqrt{\frac{9390}{150}} = \sqrt{62.6} = \pm 7.9 \text{ or } \pm 8$$

1. Determining Size Intervals or Sets: The size range of the squid as presented in the table is considerably drawn-out and space-consuming. This disadvantage is quickly eliminated by placing the individual sizes into sets with intervals of 5. (column A).

2. Determining Midpoint of Intervals: The middle size in each set. Ranges within sets are generally odd-numbered so that midpoints can be determined with ease, eliminating the involvement of fractions. (column B) Midpoint-X.

3. Frequency: The number of squid which fall into each set. (column C) N = the sum of f (Σ f).

4. Frequency x Midpoint: gives the approximate total of scores (inches in this case) in each set (column D). The sum of the frequency x midpoint is expressed as (Σ fx).

5. Determination of the Mean: Using the formula, $M=\Sigma\frac{fX}{N}$, the average size of the squid can be obtained. Specifically, the mean in this case refers to the average arithmetical size of squid (expressed in inches) of that particular sample and at that particular depth.

6. Determination of Delta (Δ) x: refers to the degree of variability, expressed as plus or minus of the midpoint from the mean. The formula for arriving at Δx is:

$\Delta x = X-M$

Delta x is an intermediate step in determining standard deviation. (column E).

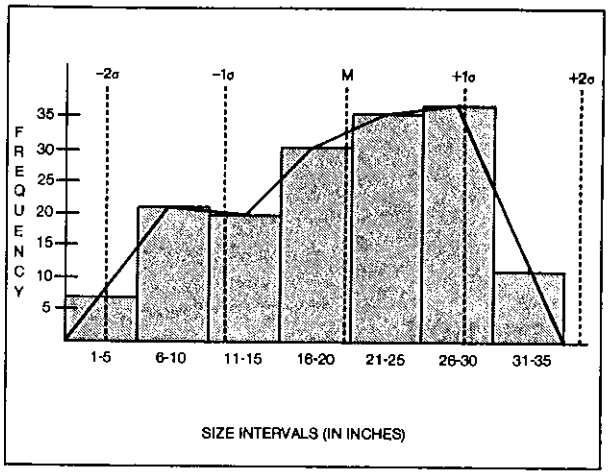
7. Determination of Delta X^2: an intermediate step in arriving at SD. Each Δx value is first squared before continuing. (column F).

8. Determination of Frequency times Δx^2: This value is obtained by multiplying the frequency within each set with the Δx^2 figure. The sum of $f\Delta x^2$ is used to determine standard deviation. (column G).

9. Determination of Standard Deviation: two values are needed at arrive at SD: $f\Delta x^2$ and N. Study the formula above.

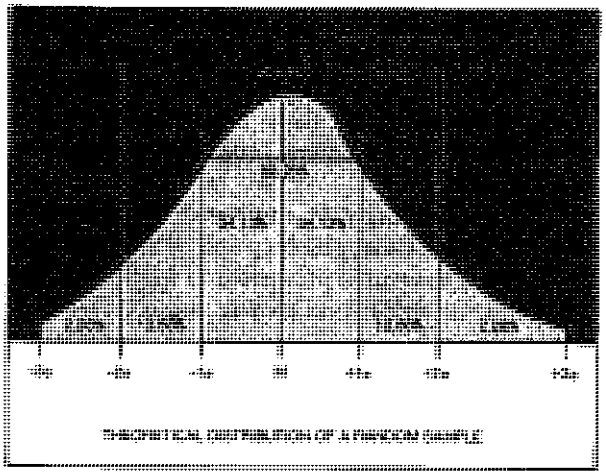
The SD$\pm$8 refers to the statistically accepted, average deviation from the mean size of squid (20 inches). The fishery biologists, then can expect a larger proportion of squid in future samples (at a depth of 25 meters, and assuming that all other factors are constant) to fall within the 12-28 size range. True? Discuss the degree of validity of this statement.

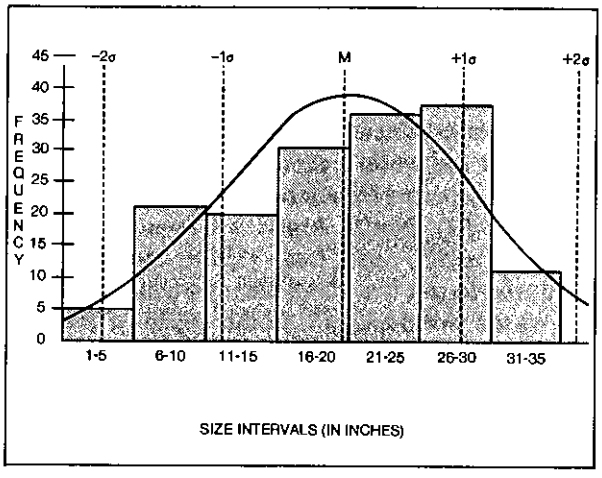
10. Plotting the Distribution of the Sample: The tendency of squid in this sample (and, possibly, in future hauls at 25 meters) to fall within the 12-28 inch size range can best be seen and understood if plotted on a histogram and superimposing a frequency polygon to clarify the resulting distribution of sizes:



The Size Distribution of 150 Squid Caught at 25 meters Depth off the Florida Coast, September, 1967.

11. Comparison of Sample Distribution with a Normal Distribution Curve: The normal distribution curve is a theoretical or ideal picture of a distribution based upon random sampling. Its use is important in statistics because it serves as a standard with which actual distributions can be compared.





A Comparison of the Theoretical Distribution Curve with the Actual Size Distribution Histogram of Squid Caught at 25 Meters Depth off the Florida Coast.

What does the above graph show? Discuss.

The plotting of the Theoretical Distribution Curve *cannot* be achieved by guessing or by merely super-imposing a bell-shaped curve on the graph. Rather, the TDC must be carefully computed for each case.

a) Determining the Height of the TDC:

$$y = \frac{N}{2.5\ (\sigma i)}$$

y = height of the TDC
N = total sample
2.5 = constant
(σi) = the number of intervals between the actual mean and one SD.

In this sample, one SD is 1.6 (1 3/5) intervals from the mean. Therefore,

$$y = \frac{N}{2.5\ (\sigma i)} = \frac{150}{2.5\ (1.6)} = 37.5$$

Using the y axis (vertical) as a guide, mark the height of the TDC on the actual mean line.

b) Determining the Height of the TDC Above and Below the Mean: Once the height of the TDC on the mean line has been calculated, the remainder of the curve can be computed as follows:

Height of the CURVE	1/2 SD = 88.3% of y
above and below the	at 1 SD = 60.7% of y
mean	1 1/2 SD = 32.5% of y
	2 SD = 13.5% of y

For this sample, then the relative heights of the TDC, above and below the mean, were:

1/2	SD = 33.11
1	SD = 22.76
1 1/2	SD = 12.18
2	SD = 5.05

Using the y axis as a guide, mark the heights of the TDC at the designated points. Carefully connect all of the points. What difficulty arises when drawing in the top of the curve?

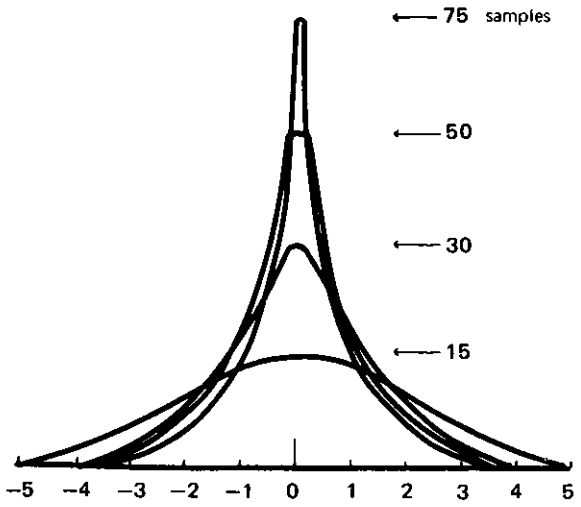
CONCLUSION

Two of the most important considerations in collecting data are the sampling technique and the sample size. The reliability of statistical analysis depends largely on the limitations of the single bit of tests performed.

All other things being equal, the Gaussian distribution curve will appear normal regardless of the number of tests performed. The average value of a group of tests will be closer to the mean than will the value of a single test.

The larger the sample the closer the average will agree with the mean or, the larger the sample the smaller the standard deviation (σ).

The diagram illustrates the tendency toward the mean as the sample size increases. The distribution retains the normal "bell-shaped curve," but its standard deviation decreases as the square root of the number of tests in the sample.



For general biological procedures, a minimum of 50 tests is considered necessary to obtain a valid mean value and workable standard deviation. Below 50 tests the standard deviation increase is fairly large while from 50 to 75 tests the decrease is relatively small.

CONSIDERATIONS AND LIMITATIONS IN USING STATISTICAL METHODS

1. Size of the sample.
2. Specific area in which sampling occurred.
3. Defining conditions under which sampling occurred. (Season, temperature, salinity, type of sampling device, etc.)
4. How accurate is the data collected?

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A STATISTICAL ANALYSIS OF A FIDDLER CRAB COLONY

TO THE TEACHER

This study was written with the assumption that the required samples of crabs will be provided for the student. It would be easy to alter this plan and involve students in the field collecting phase. Allow them to devise their own technique for random sampling of a local colony. Tide (low tide is the best time), size of colony and degree of sampling above and inside burrows are pertinent considerations.

The most important outcome of this exercise should be: a) An awareness of the scientific value and limitations of random sampling. b) Practical experience in applying this technique. c) Applying statistical methods to an actual biological problem.

TO THE STUDENT

Fiddler crabs are social arthropods. They are found in colonies ranging from less than 100 to many thousands.

Three species of fiddler crabs, of the genus *Uca*, are native to Florida and occur along both the Atlantic and Gulf coasts. Since one claw is greatly oversized, males of the genus are easy to identify. They are frequently observed waving this huge appendage. The outstanding behavioral characteristic of fiddler crabs is the rhythmic elevation and lowering of the oversized claw. The significance of this gesture has been described by various workers as non-sexually territorial, sexually territorial, a sex attractant, a challenge to other males and as various combinations of all these possibilities. (Crane, 1957)

PURPOSE

To statistically analyze the fiddler crab colony at (location) on the basis of a random sample.

MATERIALS

Small aquarium
Metric ruler
Balance
Beach sand
Saline water (less than 20 ppt)

PROCEDURE

Collecting and Keeping

Fiddler crabs are found in salt marshes, at the intertidal edge of the water. Different species prefer different substrates, usually either sand or mud. Collecting is easiest at low tide.

One collecting method requires a bucket with a ramp leading up to it, generally of plywood with low sides to prevent crabs from falling off the edges. The crabs are driven up the ramp and fall into the bucket. Another method requires a pencil-sized twig. The crabs which retreat into holes generally don't go very deep; the twig is pushed through the sand or mud at an angle to intercept the hole below the crab, which is then quickly dug from its burrow.

The crabs in the classroom don't need to burrow, but do need clean water and food available to them. The water should be taken from the collecting location, and replenished with fresh water. If the crabs are to be kept more than one week, a complete change of water each week with new brackish water from an area similar to the collecting site is advisable. The crabs do well on dry dog food.

1. Each class will be responsible for the well-being and measurement of 50 crabs.
2. Observe and measure each of the following characteristics;
    a. sex
    b. length of chela (the larger claw if a male; either claw if a female) in mm
    c. mass of crab in grams
3. Record your data for each specimen on the chart below.
4. Return the crabs to the storage tank . . . alive!

STATISTICAL ANALYSIS

On the basis of class totals calculate and graph, if required:

1. Distribution of sex in the colony.
2. Mean length and standard deviation (SD) of male and female chelae.
3. Mean mass and SD of males and females.
4. Frequency distribution of mass and length.
5. Number of right-clawed and left-clawed males. What percentage of the males were right-clawed?

QUESTIONS FOR CONSIDERATION

1. What is your opinion of the accuracy of the method described to sample the colony? If needed, how could you improve upon it?
2. What are some other possible limitations of this study?
3. How does the theoretical distribution of sex (50-50?) compare with the actual distribution?

MALE			FEMALE		
SPECIMEN #	LENGTH OF CHELA	MASS	SPECIMEN #	LENGTH OF CHELA	MASS

4. Is there any correlation between:
   a. sex and mass?
   b. lengths and masses?

5. On the basis of your statistical analysis and after carefully considering the limitations of this study, what projection(s) can be made concerning the sex distribution, mass and chelae-length characteristics of the colony under study?

6. Aside from studying sex distribution and mass-length relationships, which other characteristics of fiddler crabs could be analyzed?

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SEA URCHIN FERTILIZATION AND DEVELOPMENT

TO THE TEACHER

Sea urchins are an excellent organism for use in illustrating the process of reproduction, including: (a) morphological structure of sperm and egg; (b) fusion of gametes; (c) development of the fertilized egg.

Two species of sea urchins are recommended for use as laboratory animals, *Arbacia punctulata* and *Lytechinus variegatus.* These are relatively common in occurrence, easy to collect, hardy in the uncrowded aquarium and possess ripe eggs and sperm from September to June (probably best in February, March, and April).

There is no external feature that can be used to separate the sexes in the field, therefore, it is recommended that 10-12 specimens be collected to insure obtaining at least one member of each sex. To insure the collection of mature specimens, select only those specimens with a test (shell) diameter of from 6-8 cm. In areas where live sea urchins are unavailable or where local population are small in size, it is recommended that "Embryology of Live Bearing Fishes" be used.

The instructor should prepare a fresh supply of eggs and sperm each hour. This demonstrates the method of obtaining gametes to the class and insures an uncontaminated supply.

An electrical method for determining sex (E.B. Harvey 1952, 1953, 1954) is recommended here for the collection of eggs and sperm to be used in this unit. This method is done without harming the animal and the same animal can be used repeatedly.

The eggs or sperm are obtained by passing an alternating current of 10 volts through the animal. Regular 60-cycle, 110 volt current can be used by reducing the voltage to 10 volts with a transformer. Lead electrodes have been found best. Place the animal aboral side up covered with sea water. The electrodes are placed at two points on the animal's shell. The eggs or sperm will extrude from the five gonopores, sperm-thin white, eggs-thicker red or orange in *Lytechinus.* The shedding ceases when the current stops.

Another method described below involves the dissection of the animal and is only recommended when a study of the internal anatomy is also desired.

The urchin is washed in cold running tap water for two or three minutes to remove detritus and eggs or sperm that may have collected on its test, then rinsed in fresh filtered sea water.

Place the animal with its oral side up (inverted from its normal position) and cut the test along the area of its greatest circumference.

The oral portion of the test is now discarded and the aboral is rinsed with filtered sea water.

It is possible to identify the sex in some urchins. The female gonad (ovary) is red or orange colored and the male gonad (testis) is white.

Remove the female gonads using a spoon and place them in a petri dish or finger bowl of fresh filtered sea water and allow to stand five to ten minutes. Ripe gonads will immediately begin to eject gametes. The eggs will appear as yellow spheres and sperm as a milky fluid. Filter the eggs through cheese cloth to remove detritus and pieces of tissue. Place the eggs in 250-500 ml of fresh filtered sea water. They will remain healthy for one to two hours. The eggs will settle to the bottom of the container. They can be removed with a pipette as needed.

The testes can be stored in a petri dish without water in a refrigerator (8°C) for four or five days and remain healthy. Sperm become active when placed in sea water, but die after approximately one hour.

After each urchin has been opened, all instruments (including your hands) must be washed thoroughly. This will prevent contamination of subsequent specimens.

Prepare the sperm and egg suspension for the class by adding one drop of sperm to 1 ml of eggs in 250 ml of fresh filtered sea water. This should be sufficient for 25-30 students.

A second method is to place 1 ml of eggs in a petri dish and add sperm with a toothpick using only the amount of sperm picked up by the end of the toothpick. Stir the solution gently with the toothpick to insure even distribution.

One to two hours are required for completion of this exercise.

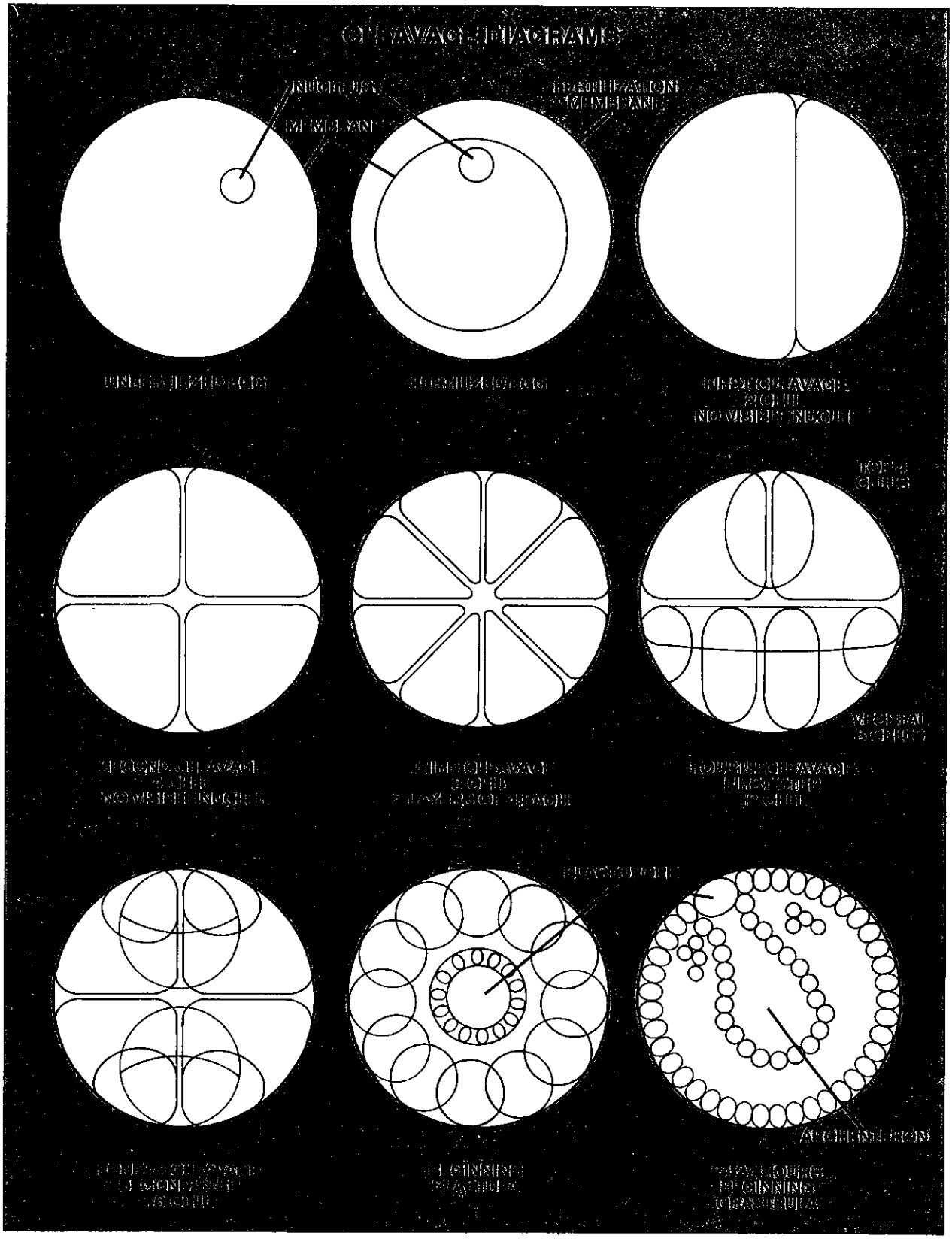
TO THE STUDENT

Sea urchins are mentioned in the writings of many of the ancient Greeks and Romans as food. Urchin eggs are still considered a delicacy in many parts of the world. Some brave souls in the class may wish to taste this particular brand of "caviar."

The ancients, according to Pliny (23-79 A.D.), used sea urchins as medicine. The urchin was ground, spines and all, mixed with a cup of wine or vinegar, and quaffed. In some cases, urchins were burned with snake skins and frogs, the ashes were mixed with vinegar. One cup a day was drunk to improve eyesight.

This exercise illustrates the principles of fertilization and development of the egg.

Sea urchins normally discharge gametes into the sea as the water warms in the spring. This process, external fertilization is very common along aquatic organisms.


CLEAVAGE DIAGRAMS
NUCLEUS
MEMBRANE
FERTILIZATION MEMBRANE
TOP & CUES
VEGETAL HEMISPHERE

After a single spermatozoan has penetrated the egg, a fertilization cone is formed which may last five minutes. Secondly, a fertilization membrane rises over the cone and covers the entire egg in a few seconds. The egg now elongates and the first cleavages begin, leading to the ultimate formation of the free swimming pluteus.

PURPOSE

To observe the development of a zygote during its early stages of division.

MATERIALS

Microscope
Sea urchins
Petri dishes
Watch glasses
Slides (plain, concave, or blister)
Cover slips
Fresh sea water (filtered)
Pipette
Small pieces of glass tubing
Cheese cloth
Vaseline
Scissors

PROCEDURE

1. Secure a drop of the sperm and egg suspension. Place on a hanging drop slide (sealed in Vaseline), a concave slide (cover slip sealed in Vaseline), or a blister slide.
2. Record observations made under the microscope. Use 10 x objective.
3. After completing observations, place the slide on two glass rods in a petri dish, add water to cover ½ of the rod but not the slide. This will provide a moist incubation chamber and will possibly allow observations to continue up to 48 hours.
4. Repeat microscopic observations as often as possible during the day.

ORGANIZED DATA RECORDING

1. Measure size of unfertilized egg, sperm, and initial fertilized egg. (See "Measuring with a Microscope.")
2. Record the time lapse between the mixing sperm and egg to formation of:
   a. Fertilization cone
   b. Fertilization membrane
   c. First cleavage
   d. Second cleavage
   h. Third cleavage
   f. Fourth cleavage
   g. Blastula
   h. Gastrula
   i. Pluteus
3. Record observed physical changes that occur with stages listed above.

QUESTIONS FOR CONSIDERATION

1. How is development of the sea urchin zygote similar to development in mammals?
2. In what direction does cleavage occur in each step of division?
3. Compare the motility of egg and sperm.
4. How do you explain "parthenogenetic" development in sea urchin eggs?
5. What are the advantages and disadvantages of liberating sperm and eggs into the oceans?

LIMITATIONS AND SOURCES OF ERROR

Use only freshly obtained animals or any organisms which give comparable results.

Fresh, well aerated sea water is essential.

Evaporation of sea water from observation chamber raises salinity, must be avoided.

Clean glassware must be used.

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EMBRYOLOGY OF LIVE BEARING FISHES

INTRODUCTION

Large numbers of brackish water and fresh water fishes utilize the process of internal fertilization made possible by the modification of part of the anal fin into a copulating organ in the male. Among these ovoviviparous fish, the female retains the eggs in the ovary during the embryonic development period. At birth the fry are ready to care for themselves, indicating that a great amount of development occurs before birth.

TO THE TEACHER

This exercise requires one laboratory period for Part A and two laboratory periods for Part B.

If sufficient student interest is developed, they may wish to explore courting behavior, population counts, or sexual dimorphism.

The fish to be used include: *Gambusia affinis* (Mosquito Fish); *Heterandria formosa* (Dwarf Top Minnow); and *Mollienisia sp.* (Sailfin Molly). These can be obtained by seining fresh to brackish water, ditches, pools, and ponds. If these are not available *Lebistes reticulatus* (Guppy) will suffice.

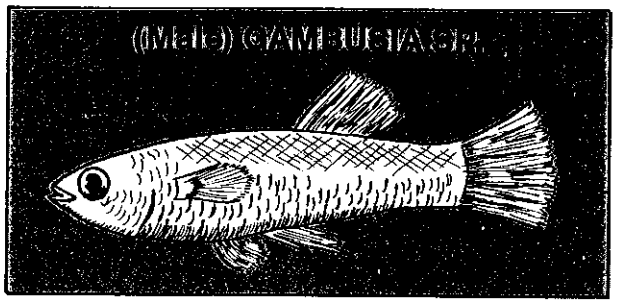
Each student must dissect several gravid females in order to obtain a large range of embryonic stages.

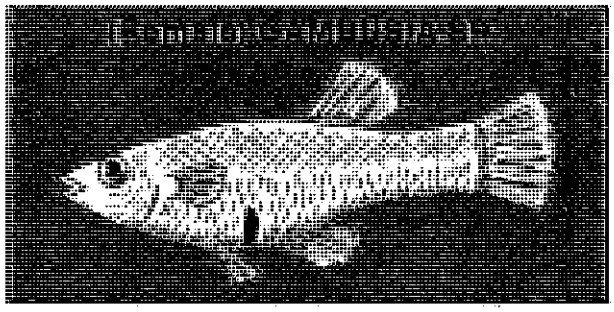
The mature gravid female of the *Gambusia sp.* is easily recognized by its swollen abdomen and large dark pigmented spot. (See illustration.)

A supply of specimens (5 per student) should be on hand before beginning.

TO THE STUDENT

The mature female *Gambusia sp.* is gravid almost continuously. Secondly, the female of the species is usually larger than the male and more numerous. The female of some fish species such as *Gambusia sp.* or *Lebistes reticulatus* possess a dark pregnancy spot. In choosing your specimens





for dissection, select fish with spots of varying size and intensity of pigmentation.

The female *Gambusia* exhibits superfetation (the possession of more than one set of embryos at a time) by retaining sperm from the initial mating and producing a second set of eggs while the first is developing. These sperm are used to fertilize the second set of eggs during the development of the first set. The development of the second set of eggs is retarded until the birth of fry, then development begins rapidly.

The average time required for the development of the egg until fry are produced is 28 days.

The ovoviviparous fishes have internal fertilization, internal development of the fry, and live birth, but there is no placenta (the organ in most animals by which the fetus is joined to the uterus and is nourished). The fertilized egg develops without benefit of attachment to its mother; instead it depends on the yolk.

PURPOSE

To examine and determine the varying stages of embryonic development in an egg during its growth.

PART A — PRESERVED SPECIMENS

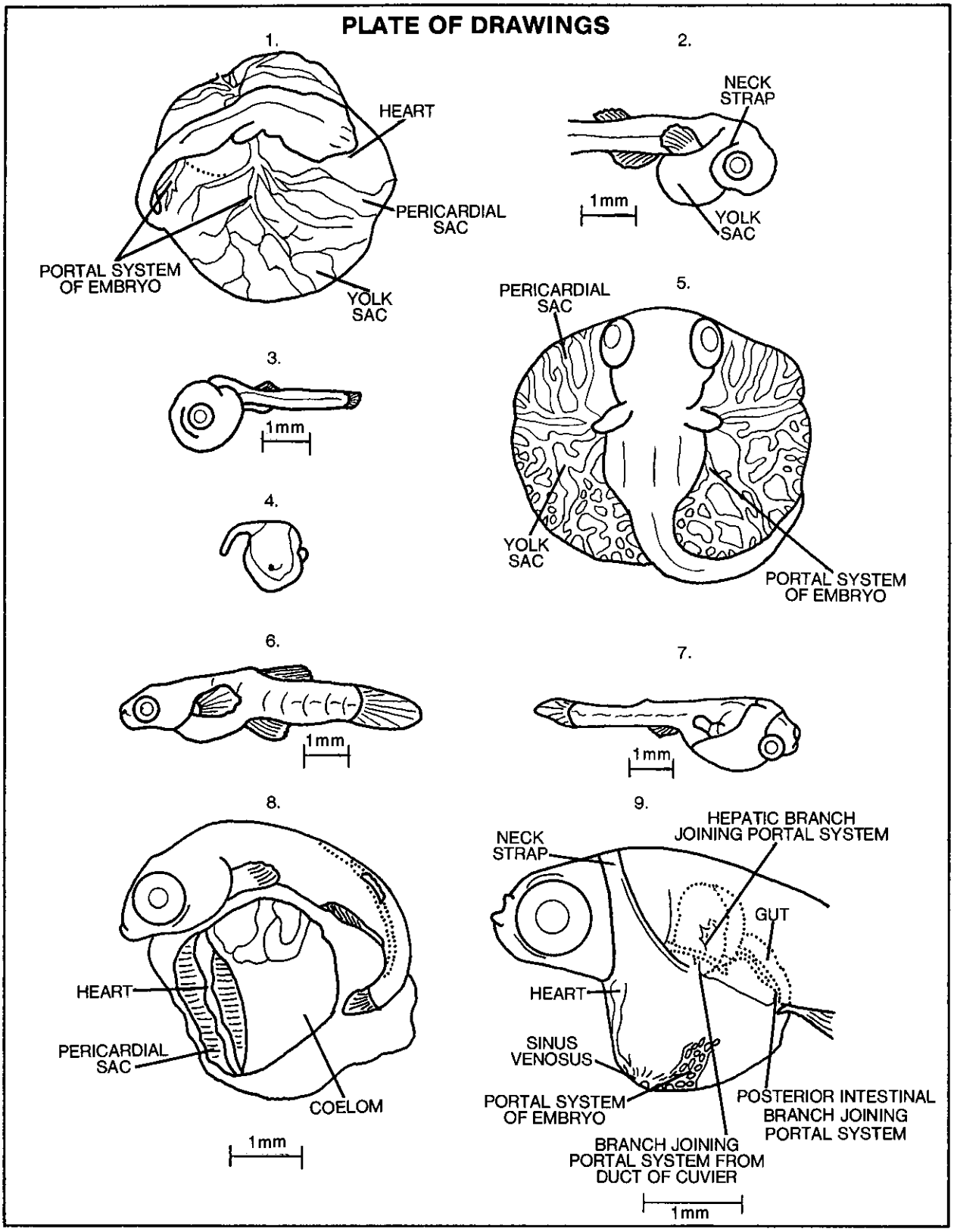
MATERIALS

5 gravid females (preserved)
Dissecting needle
Small pair of sharp scissors or razor blade
Watch glass or petri dish half
Dissecting microscope

PROCEDURE

1. Secure preserved specimens and petri dish.
2. Measure the standard length of each specimen.

PLATE OF DRAWINGS



3. Hold the head between your fingers and cut the head from the body behind the gills.

4. Cut the abdominal wall back to the anal opening, thus opening the stomach cavity.

5. Place under the dissecting scope and remove the white to yellow spheres observed in the viscera. These will be the embryos and eggs.

6. Sort these spheres into groups according to the amount of development observed.

Key to description:

1. Seven-day embryo of *Fundulus heteroclitus:* To illustrate spreading of portal system to pericardial sac.

2. 5.5 mm embryo of *Gambusia affinis:* To illustrate typical neck strap structure.

3. 3.5 mm embryo of *Heterandria formosa.*

4. Very young embryo of *Heterandria formosa.* Head is enveloped by the external and internal folds of pericardial sac.

5. Nine-day embryo of *Fundulus heteroclitus* somewhat flattened dorso-ventrally.

6. Embryo of *Heterandria formosa* at close of the period of gestation. Neck strap has been absorbed and belly sac is shrinking.

7. 5 mm embryo of *Heterandria formosa.* Head has pushed through the folds of the pericardial sac leaving neck strap dorsally.

8. Diagram of a dissection of 5 mm embryo of *Poecilistes sp.* Portal network covering belly sac is not indicated.

9. Diagram of the portal system covering the yolk sac in a 7.5 mm embryo of *Lebistes reticulatus.* Blood emerges from the body of the embryo through branched arteries via the liver, duct of Cuvier, and region just in front of the anus, traverses the portal network and is received in the sinus venosus.

The abbreviations, illustrations and descriptions are adapted from Turner (1940a).

PART B — LIVE SPECIMENS

MATERIALS

Live fish

Physiological saline (0.7% solution of NaCl in distilled water)

Items listed under materials in Part A.

PROCEDURE

1. Instructions 1-5 are the same as in Part A.

2. As the embryos are removed from the viscera they are to be placed in physiological saline solution and maintained at room temperature for three to four days and observed at regular intervals. Time alive should be recorded.

3. If the embryo is well-developed at the time of dissection, it is possible to place them in an aerated aquarium and observe development to maturity.

ORGANIZED DATA RECORDING

Record the number of eggs and/or embryos removed from each female and the various stages of development for the eggs and embryos. Record the number of males and females in the sample of specimens. Records of the standard length should accompany each female.

QUESTIONS FOR CONSIDERATION

1. These fish do not produce as many eggs as some others. Why?

2. Can you find any correlation between the size of the pregnancy spot and the number of eggs and embryos per female?

3. Why are not all of the eggs and embryos at the same stage of development?

4. What is the function of the placenta in animals?

5. How does the *Gambusia sp.* fry eliminate waste material before birth?

GRAPHIC ANALYSIS

1. Graph standard length against the number of eggs and embryos per female.

2. Graph the development of eggs and embryos at each level of development.

3. Calculate the percentage of females versus males in the total sample.

LIMITATIONS

Each student should have five to ten specimens available to insure a wide range of embryonic development.

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SHARK STUDY

TO THE TEACHER

In the eyes of the high school student of marine science, the shark provides a stimulation equaled by no other animal of the sea. This laboratory study is constructed so that the class can be involved for four consecutive periods (one week's work). On the other hand, any part can be used independently of the others. Depending upon time allowed, specimens available and level of interest, any or all of the separate exercises may be expanded.

Basic Outline of Study Sequence:

I Reading Research
II External Anatomy
III Internal Anatomy
IV Brain Dissection
V Shark Teeth

Local sport and commercial fishermen will help in collecting a supply of embryo sharks. Nearby professional marine laboratories, especially those engaged in routine shark studies, can also provide specimens. Be ready to respond immediately to the call of the volunteer collector, and have storage facilities ready at all times: Freezer facilities, capable of storing the embryos, are recommended. The animals can be preserved in ordinary fixative, if there are no freezer facilities.

The embryos of the brown shark *(Carcharhinus milberti)* (Muller and Henle) reach about 50 cm before birth. Ideally, this laboratory calls for two students per shark.

Collection of live adult specimens by high school students should be discouraged. Even the more docile nurse shark *Ginglymostoma cirratum* (Bonnaterre) will bite if handled improperly. (Gilbert 1964).

If there is a known local area where fossilized shark teeth can be obtained, have students collect their own shark's teeth. If a local site is not available, shark's teeth may be obtained from biological supply houses, shell shops or curio shops.

TO THE STUDENT

Sharks are chordates with a skeleton made of cartilage. The gills open to the outside of the body through slits. Fertilization is internal and, in most species, the young are born alive. Sharks are scavengers and most are flesh-eaters. Their senses of smell and hearing are very well-developed. Teeth of the shark occur in series or rows. Tooth replacement occurs throughout the life of the shark.

Sharks' attacks upon man occur with greatest frequency between 30°N and 30°S latitudes. Since their actions are so unpredictable, most experienced divers and collectors treat the shark with caution and respect.

The Office of Naval Research which has funded the SAF (Shark Attack File) discontinued doing so in 1970. During the lifespan of SAF (1958 - 1969), 107 shark atacks were recorded in Florida, an average of 10 per year. In recent years, ⅓ of the recorded worldwide annual shark attacks have occured in Florida. Most Florida "attacks" have resulted in only minor injury. Florida has a long coastline (2nd only to Alaska in the U.S.), warm water, and a large part of the population in the water, which could account for the high number of recorded shark attacks.

Our main enemy in the water is not sharks. It is sunburn, drowning, jellyfish, long spiny sea urchins and fire coral.

The following species are fairly common in Florida waters: Blacktip Shark, Bonnethead Shark, Brown or Sandbar Shark, Bull Shark, Dusky Shark, Hammerhead Shark, Lemon Shark, Nurse Shark, Tiger Shark, Spinner Shark, Florida Dogfish, Spiny Dogfish and Sand Tiger.

THE PROBLEM

1. Through reading research, to become acquainted with taxonomy of sharks; to learn about the feeding habits of sharks: and to become familiar with present-day commercial and research interests in sharks.
2. To measure and record the external features of a shark embryo.
3. To learn about the gross internal anatomical features of a shark embryo.
4. To learn about the gross anatomy of the brain of a shark embryo.

MATERIALS

Shark embryo
Dissecting board
Newspapers and paper towels
Dissecting kit
Scales
Chart
Calipers and ruler
Anatomical Charts

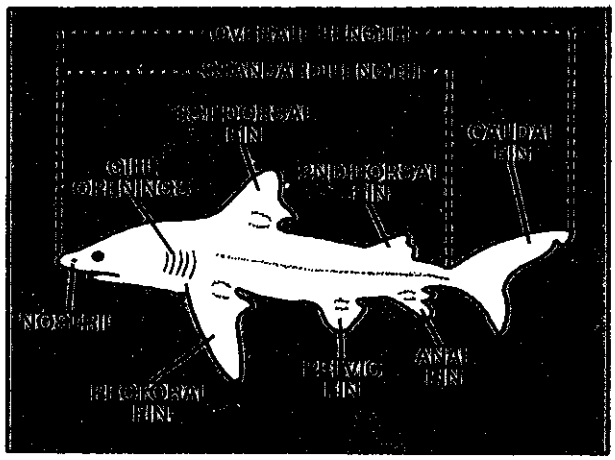
PROCEDURE

Part I: Reading Research

A. Complete the classification of your specimen:
Phylum: _____
Class: _____
Subclass: _____
Order: _____
Suborder: _____
Family: _____
Scientific Name: _____
Genus Species

B. Answer the following questions. State the source(s) of your reference(s).
1. List the physical characteristics which separate the sharks from the bony fishes (Class Osteichthyes).
2. What are the feeding habits of sharks?
3. Of what economic importance are sharks?
4. Of what research interest are sharks? Where is reseach on sharks being carried on in Florida?
5. Under what conditions are sharks most dangerous to man?
6. What other animals are in the same class category as sharks?
7. What species of sharks are found in the coastal waters nearest the school?

EXTERNAL ANATOMY OF A TYPICAL SHARK



PART II: External Observations

Record the following data:

_____ _____
Common Name Scientific Name
Length (overall) _____ cm
Length (standard) _____ cm
Width: at head (A-A) _____ cm
at middle (B-B) _____ cm
at 2nd Dorsal Fin (C-C) _____ cm
Mass _____ gm (454 gm = 1 lb.; 28 gm = 1 oz.)

Fins: (number and description)
_____ 1st Dorsal _____
_____ 2nd Dorsal _____
_____ Pelvic _____
_____ Anal _____
_____ Caudal _____
How many gill slits? _____
Are gill opercula present? _____
Where is the cloacal opening located? _____
Are claspers present? _____. If so where are they located? _____
Describe the surface color of specimen _____
_____
Describe the texture of the skin surface _____
_____
Open the mouth of the animal. Do you see any teeth? _____
With the mouth held open, feel for the presence of teeth with a fingertip. Record findings:
Are teeth present on both the upper and lower jaws?
_____
Draw a side view of specimen and label the following parts: nostril, mouth, eye, gill slits, all fins including the caudel (tail) fin, location of cloacal opening, claspers (if present), lateral line, spiracle. Significant external skin surface patterns should be shown. Indicate with lines "A-A", "B-B" and "C-C" where width measurements were taken.

PART III: Internal Observations

Expose the viscera by making a median longitudinal incision, ventrally, beginning at the cloacal opening and proceeding anteriorly. Describe the liver (size, number of lobes, texture and color). _____
_____

Remove the liver. How much does it weigh? _____ gm. What percentage of the total body mass (weight) consists of liver alone? _____%. Is the alimentary canal longer than the body (exclude tail)? _____. Locate and name the portions of the alimentary canal, beginning with the mouth:

Remove the entire digestive tract. Examine, then slit the small intestine. Is a "scroll" present in this animal? _____
Locate the following organs. Indicate with a check (✓) if the organ(s) is (are) present in the specimen. Tell where they are located.

gall bladder	_____	_____
pancreas	_____	_____
spleen	_____	_____
kidneys	_____	_____
ovary (ovaries)	_____	_____
testes	_____	_____
rectal gland	_____	_____

Locate the heart, Is there a partition separating the heart from the other organs?
Length overall _____ cm
Width overall _____ cm

Since the heart of embryo sharks is rather small, additional reading research will probably be necessary to complete the following questions.

Does the heart of the living shark contain both arterial and venous blood? _____

Explain _____

_____

How many chambers are present? _____

Name them: _____

PART IV: Brain Observation

Before dissecting, prepare a sketch of the shark (or related animal) from reading research. Label the following parts: Olfactory lobe, Olfactory tract, Cerebral hemisphere, Optic lobe, Cerebellum, Medulla oblongata, Spinal cord, Cranial nerves.

Remove as much skin and tissue as possible dorsally, between the eyes and over the general cranial area. Remember that the shark has a cartilaginous skeleton. Extreme care and patience is required. Expose as much of the brain as possible. It will be difficult to remove the brain, since it is held together very delicately.

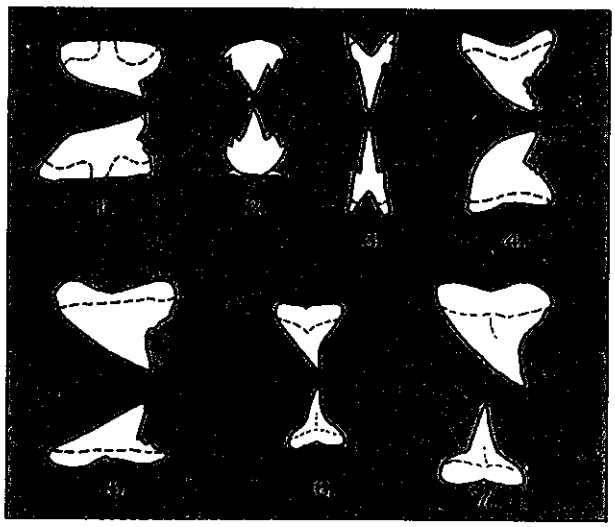
With the help of the drawing, locate as many of the labeled parts as possible. Now, prepare another sketch of the brain of the specimen. How do the drawings compare?

How many cranial nerves are there?

Does the shark eye contain a crystalline lens?

Trace the optic nerve (Cranial Nerve No. II) to the brain.

How many muscles move each eye?



An upper and lower tooth to the left of the symphasis. 1. Spined dogfish; 2. Nurse shark; 3. Sand shark; 4. Tiger shark; 5. Hammerhead shark; 6. Bull shark; 7. Sandbar shark.

PART V: Sharks' Teeth

Shark teeth have fascinated man for many years. Shell pendants and tools have been discovered in Indian "dig sites" which were drilled with sharks teeth. Inland Indian mounds have contained both recent and fossilized sharks teeth, indicating that these were valuable tools and perhaps valued jewelry. The shark tooth drill was made by notching the end of a stick and placing the tooth in the notch with the point sticking out. The stick was straight, 6" - 12" long, with a diameter slightly larger than the width of the sharks tooth.

Fossil sharks teeth can be found throughout Florida and the coasts of other Southeastern states. One method for collecting sharks teeth is sifting beach or off-shore sand through a homemade basket made from ¼ hardware cloth (welded wire). (Live sharks don't take too keenly to having their teeth pulled.)

Taxonomists (biologists who classify living organisms) and paleontologists (scientists who study fossils) find sharks teeth important in identifying shark species.

1. Using the books listed under References, with the sharks teeth that either you collected or your teacher furnished, identify the sharks. Don't get discouraged. Taxonomists consider it excellent work if they can identify as much as 30% of the teeth.

2. Make a sharks tooth drill and drill through a shell. How long did it take you? Do you understand why Indians treasured their pendants and shell tools?

QUESTIONS FOR CONSIDERATION

1. Trace the pathway of blood through the heart, gills and body of the shark.
2. What device(s) does the shark lack, making it necessary for the shark to be almost continually on the move?
3. Is the remora (Echeneis naucrates) a commensal or symbiont?
4. What is meant by (a) individual feeding pattern and (b) mob feeding pattern?
5. How do shark teeth differ between the species?
6. Can a shark be "trained"?
7. What purpose(s) does the "scroll" intestine serve?
8. Which species have a "spiral valve" intestine?
9. What purpose(s) does the lateral line serve?

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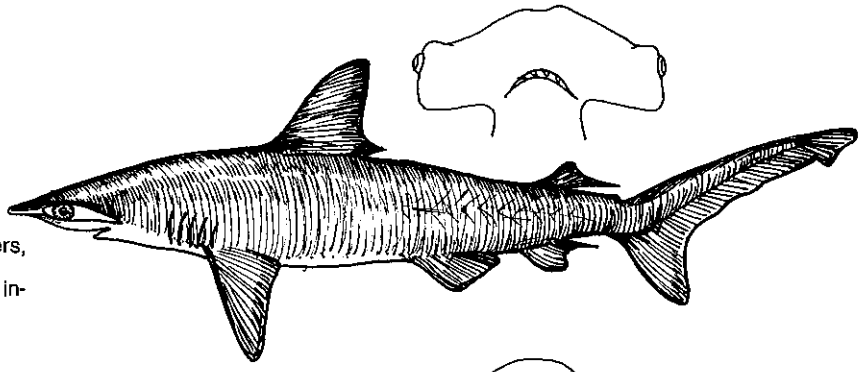
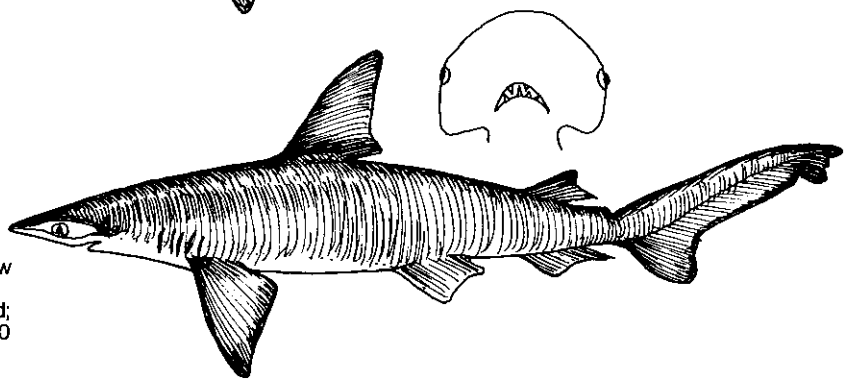
SCALLOPED HAMMERHEAD (Sphyrna lewini)

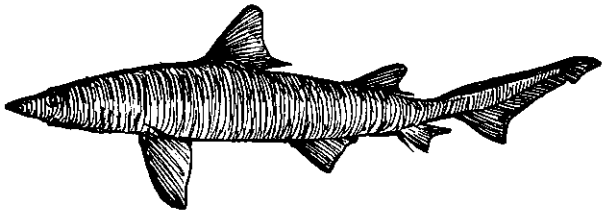
MAXIMUM LENGTH: 11 feet.
COLOR: Grayish above, paler below.
RANGE: Inshore and offshore tropical waters, Atlantic and Pacific.
REMARKS: Smooth-edged teeth; head indented at midline.



BONNETHEAD, SHOVELHEAD (Sphryna tiburo)

MAXIMUM LENGTH: 3.5 feet
COLOR: Grayish brown, lighter below.
RANGE: Tropical and subtropical shallow waters; flats, bays, passes, estuaries.
REMARKS: Rounded, shovel-shaped head; often seen in small groups of 10 to 20 individuals



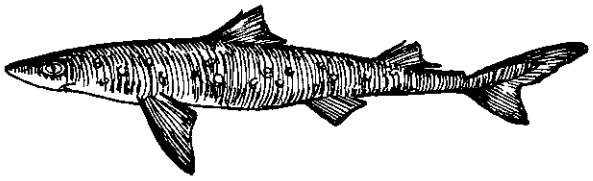


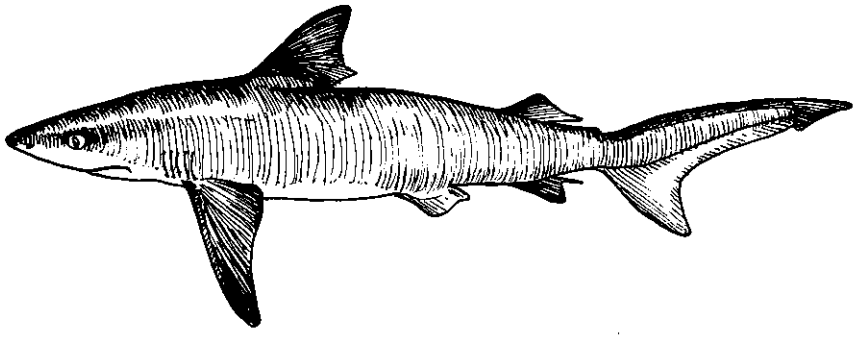
FLORIDA DOGFISH, FLORIDA SMOOTHHOUND (Mustelus norrisi)

MAXIMUM LENGTH: 3.5 feet
COLOR: Grayish to brownish above, lighter below.
RANGE: West coast of southern Florida to Florida Keys.
REMARKS: Smaller teeth and narrower mouth than M. canis.

SPINY DOGFISH, SPURDOG, PIKED DOGFISH (Squalus acanthias)

MAXIMUM LENGTH: 3 feet.
COLOR: Brownish or grayish, with irregular white spots.
RANGE: Temperate waters, worldwide.
REMARKS: No anal fin; poisonous spines on anterior edges of both dorsal fins.



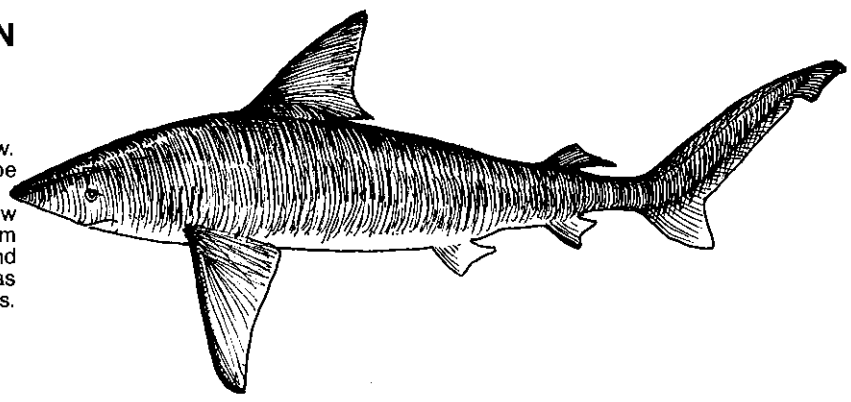


DUSKY SHARK (Carcharhinus obscurus)

MAXIMUM LENGTH: 11 feet.
COLOR: Grayish to bluish above, lighter below.
RANGE: Inshore and offshore waters, tropical and temperate Atlantic and Pacific.
REMARKS: Ridge-backed; similar to, but generally larger and more slender than C. milberti.

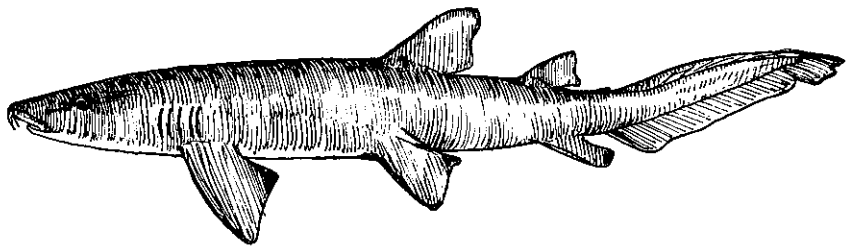
SANDBAR SHARK, BROWN SHARK (Carcharhinus milberti)

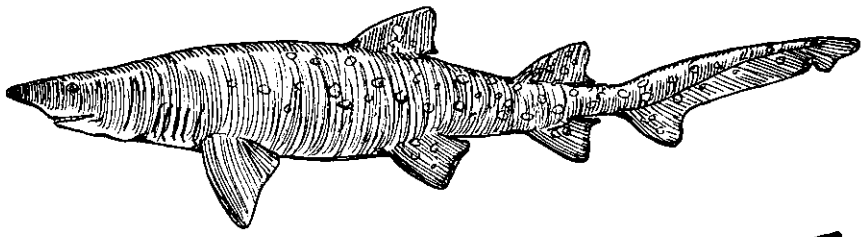
MAXIMUM LENGTH: 7.5 feet.
COLOR: Brownish gray above, lighter below.
RANGE: Inshore and offshore waters, Cape Cod to Florida.
REMARKS: Most common large shark in New York-New Jersey waters; distinguished from C. obscurus by its heavier "shoulders" and higher dorsal fin, situated further forward; has dorsal ridge; synonymous with C. plumbeus.



NURSE SHARK (Ginglymostoma cirratum)

MAXIMUM LENGTH: 14 feet; usually not more than 9 feet.
COLOR: Grayish to yellowish brown; younger specimens often have irregular dark spots.
RANGE: Western Atlantic, Rhode Island to Brazil.
REMARKS: Small eyes; prominent nasal barbels; fourth and fifth gill slits very close together.



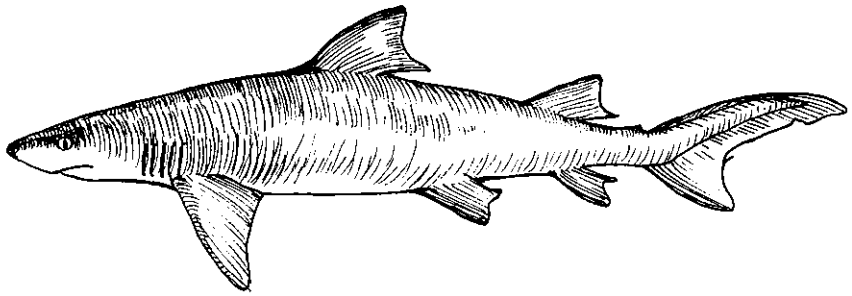


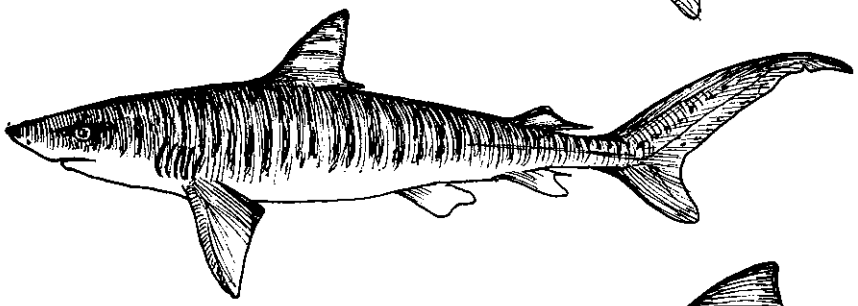
SAND SHARK, SAND TIGER (Odontaspis taurus)

MAXIMUM LENGTH: 10 feet.
COLOR: Gray-brown with irregular dark spots.
RANGE: Western Atlantic; genus represented worldwide by closely allied species.
REMARKS: Pointed snout with protruding snaggle teeth; light-colored eyes; two large dorsal fins.

LEMON SHARK (Negaprion brevirostris)

MAXIMUM LENGTH: 11 feet.
COLOR: Yellowish brown above, lighter below.
RANGE: Inshore in the western Atlantic.
REMARKS: Short, wide snout; dorsal fins of almost equal size.



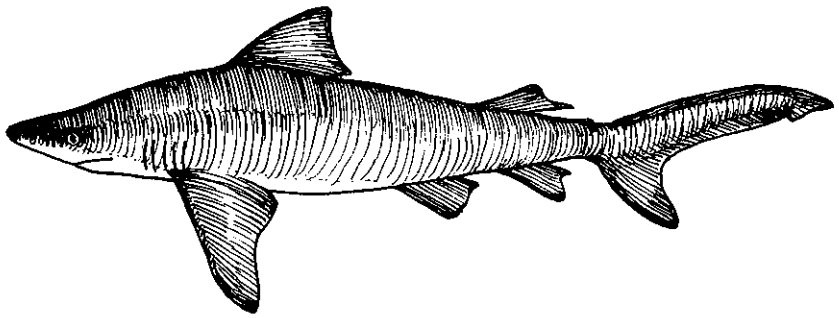


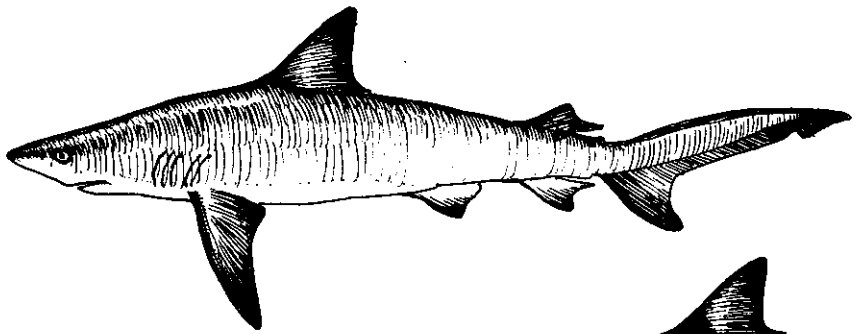
TIGER SHARK (Galeocerdo cuvieri)

MAXIMUM LENGTH: 20+ feet.
COLOR: Gray to ochre, marked with transverse bands that fade with age.
RANGE: Tropical waters, worldwide.
REMARKS: Squarish snout; pronounced labial fold on upper lip; teeth uniquely cockscomb-shaped; long upper tail lobe.

BULL SHARK, CUB SHARK (Carcharhinus leucas)

MAXIMUM LENGTH: 10 feet.
COLOR: Grayish above, lighter below.
RANGE: Worldwide inshore waters, including many freshwater lakes and rivers.
REMARKS: Lake Nicaragua shark, Ganges River shark, Zambezi shark, and other widely distributed species have all been identified as C. leucas; no dorsal ridge, but often trematode parasites or scars between dorsal fins.



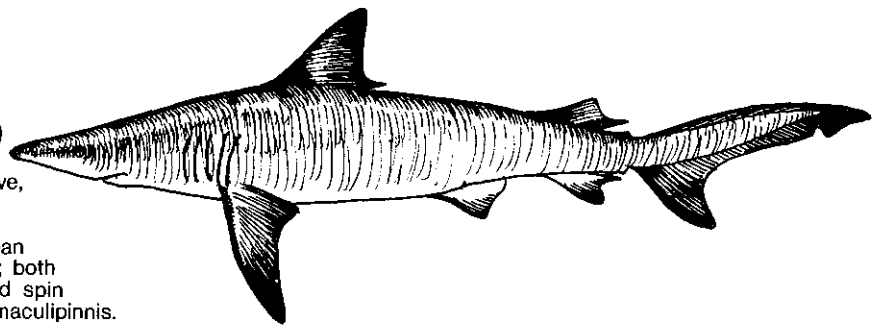


BLACKTIP SHARK (Carcharhinus limbatus)

MAXIMUM LENGTH: 7 feet.
COLOR: Dark gray, bronze, or slate blue above, lighter below.
RANGE: New York to Brazil in the Altantic; Lower California to Peru in the Pacific.
REMARKS: No dorsal ridge; black-tipped fins.

SPINNER SHARK (Carcharhinus brevipinna)

MAXIMUM LENGTH: 8 feet.
COLOR: Dark gray to bronze or bluish above, lighter below.
RANGE: Tropical and subtropical Atlantic.
REMARKS: No dorsal ridge; smaller eyes than C. limbatus, which it strongly resembles; both species known to leap from water and spin before reentering; synonymous with C. maculipinnis.



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FIN RAY AND VERTEBRAE ANALYSIS OF BONY FISH

INTRODUCTION

Most of the available taxonomic keys to the fishes use such characteristics as the number of spines and rays in the dorsal and anal fins and the number of transverse rows of scales crossing the lateral line to separate species. It is difficult to find a color plate that illustrates the exact specimen on hand. This is especially true of the small brackish water and freshwater fishes of the Southeastern United States.

TO THE TEACHER

The method of staining the fish skeleton used here works well for small fish. By partially fleshing the skeleton, larger specimens can be stained successfully. The embryo of fish can be placed in a plastic bag with fine holes punched in it and stained for fin and vertabrae development.

One must realize that a small or juvenile fish has a larger head and eye in proportion to the body than does the adult specimen. The number of fin rays and scale rows remains constant at all age levels.

It would be of great benefit to the student's understanding if preserved specimens of several months' standing were on hand to illustrate the color change brought about by preservatives when compared to live specimens in the aquarium.

Seining in shallow brackish water should produce several different good specimens such as *Fundulus sp.* (killifishes), *Gambusia sp,* gobies, blennies, pipefishes and others. The species of fish will vary with season and location sampled.

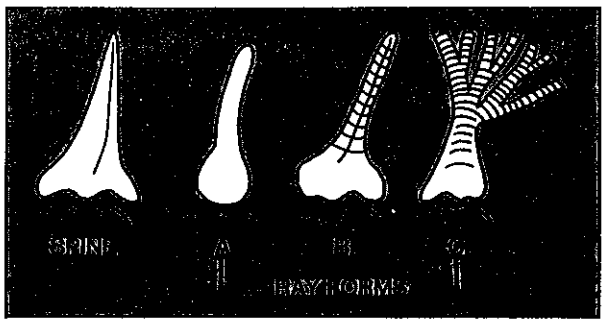
It is advised that wooden racks or trays be used for the containers. This will eliminate spillage of caustic material (KOH) and stain. It will also ensure that containers of solutions will not be shuffled out of sequence. Take special care to avoid contact with the skin of any chemicals used in this lab.

TO THE STUDENT

Using the color of fishes is not a reliable method for accurate identification because a large number of fish have the ability to modify their patterns. Secondly, others tend to fade and lose their color upon death and/or preservation. The dolphin, with its brilliant blue-green and yellow flashing in the ocean, becomes a dull grey on the dock. A large percentage of museum specimens tend to become bleached and colorless. The identification tag becomes the only clue remaining that the specimen was once a living animal.

This lab will give an adequate method of counting the number of rays present in the doral and anal fins of small fishes. These rays are frequently very difficult for the untrained eye to separate and count; this fact makes most available keys useless.

SPINES AND SOFT RAYS

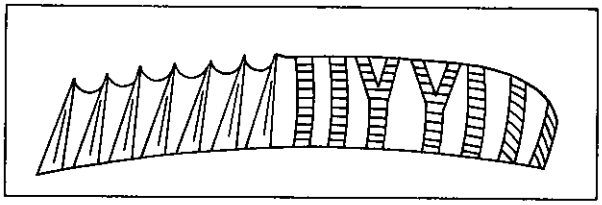


The spines are smooth, pointed, solid, and not divided into segments. Soft rays are smooth, soft and not pointed, and may occur in three forms:

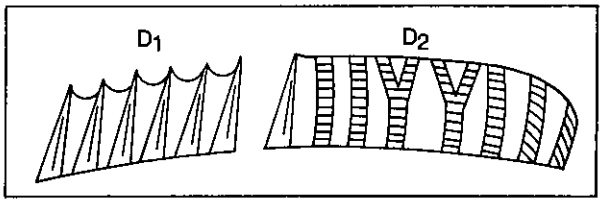
a) unbranched and non-segmented
b) unbranched and segmented
c) branched and segmented

(after Sterba [altered from Gunther] 1962)

A Roman numeral is used to represent the total spine number and an Arabic number to represent the total ray number.



In the illustration above, the code would be VII, 8. The number VII refers to seven spines; the number 8 refers to eight rays (count the base of the rays).



Here the code would be VII-I, 8. The VII represents the seven spines in D_I. The dash indicates that there are two fins, 1, 8 indicates that the second fin (D_2) has one spine and eight

rays. Thus the fin is in two parts: One part has seven spines: the other part has one spine and eight rays.

The numbers are frequently preceded by a capital letter to indicate which fin is described (example: D=dorsal).

PURPOSE

To achieve an understanding of the taxonomic principles used in cataloging fishes and to analyze the skeletal structure of fishes.

MATERIALS

Compound microscope
Several species of fish
Petri dish
White tray or a baby food jar
Alizarin Red S Stain
Acetic acid 5 ml
Glycerine 10 ml
Choral Hydrate (1% solution) - 60 ml

Saturate above solution with Alizarin Red S Stain until no more stain will go into solution. Add 2 cc of this solution to 100 ml of 4% KOH to produce the specimen staining solution.

Clearing Solution No. 1
Glycerine - 20 ml
*4% KOH - 3 ml
Distilled water - 77 ml

Clearing Solution No. 2
Glycerine - 50 ml
4% KOH - 3 ml
Distilled water - 47 ml

Clearing Solution No. 3
Glycerine - 75 ml
Distilled water - 25 ml
4% KOH

*Dissolve 4 grams Potassium hydroxide in 20 ml of distilled water then add distilled water to make 100 ml of solution.

PROCEDURE

1. Select two specimens each of 3 varieties of brackish water fishes.
2. If the specimens are between two and four inches in length, gently remove the scales.
3. Place the fishes in a white tray or enclose in a bottle with 4% KOH for bleaching. The KOH volume must cover the specimens used.
4. The specimens must be left two to three days in the sun or until completely bleached (translucent). Caution: If left too long, the specimen will tend to fall apart.
5. Immerse the fish in a container of the Alizarin Red S stain solution for approximately 24 hours or until the bones become a deep reddish-purple.
6. Place specimens in a beaker of fresh 4% KOH overnight to begin clearing process, then put fish in Clearing Solution No. 1 for two or three days.
7. Transfer specimens to Clearing Solution No. 2 for two or three days (longer if the tissue becomes more transparent).
8. Place fish in Clearing Solution No. 3 for two days.
9. Change specimens to pure glycerine for two days as the specimens are now reaching maximum transparency. In fact, they can now be stored in glycerine. Add a crystal of thymol to prevent fungus growth.
10. The fish are now placed in a petri dish half and observed under the compound microscope.

ORGANIZED DATA RECORDING

Record for each specimen:
a. number of vertebrae
b. number of fin rays in the dorsal fin
c. number of fin rays in the anal fin

QUESTIONS FOR CONSIDERATION

1. Do all specimens of each type used possess the same number of anal and dorsal fin rays?
2. What physical functions do the fins have?
3. What differences occur within the skeletal structures that separate one species from another species in your sample?
4. What other anatomical and/or biochemical characteristics could be used to classify fishes?

LIMITATIONS AND SOURCES OF ERROR

a. Time requirement the transfer of specimens requires 5 to 10 minutes each day, thus disrupting lecture time; specimens need to be close at hand to prevent lost motion.
b. Overstaining and failure to clear the specimen properly.

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STOMACH ANALYSIS OF FISHES

TO THE TEACHER

This exercise involves one field trip (fishing trip) plus one to two periods in the laboratory. With proper consideration given to planning, this can be a fun lab as well as instructional. Due consideration should be given to the type of bait used in fishing, whether artificial, live or frozen, in analyzing results.

If a fishing trip is not feasible, a seine haul along a shallow beach or estuary will provide the necessary specimens. If all else fails, go to a local fish house and secure fresh specimens.

This is a good to time to collect scales, save small specimens for staining or build up the school museum.

TO THE STUDENT

Many species of fishes make use of a wide variety of food and some authorities state that fish will eat whatever is available. Lagler et al (1962) said, "As for the manner of feeding, only one broad common characteristic prevails — the food is taken into the mouth." However, each type of fish has some preference for food just as most people prefer steak to weiners. Some species of fish have adapted to such an extent that their mouth parts have changed to accept certain food or the stomach has evolved as a curved organ, or a grinding organ similar to the gizzard of a bird. *Hippocampus* sp.; *Syngnathoides* sp. and *Aeolisus* sp. are examples of a rather odd group which are stomachless. They swallow whole prey directly into the intestine. (Brown 1957).

The adaptations of the teeth to type of food consumed is apparent in many cases. The drum and sheepshead have large blunt crushing teeth for mollusks. Sharks, bluefish, and sea trout have sharp, inwardly-curving teeth for capture and retention of smaller fish. The herbivorous fish such as mullet have oral teeth, pharyngeal teeth, or a pyloric gizzard.

The exercise will attempt to correlate the type of teeth with the stomach contents. Careful attention should be paid to the possibility of finding fish tags on the specimens caught or in the stomachs analyzed.

PURPOSE

Examination of stomach contents and tooth shape to determine food selection preference.

MATERIALS

A. Field
- Balance
- Metric rule
- Preservative*
- Field notebook
- Thermometer
- Taxonomic keys
- Syringe & needle
- Fishing equipment & bait
- Assorted plastic bottles & jars
- Labels
- Knife
- Watch
- Sewing thread

*1 part 40% Formaldehyde, 7 parts sea water

B. Laboratory
- Dissecting microscope
- Compound microscope
- Slides
- Cover slips
- Taxonomic keys
- Dissecting instruments

PROCEDURE

A. In the field

1. On reaching the site chosen, each student is to record:
   - time of day
   - tide conditions
   - cloud cover
   - wind velocity
   - type of bottom
   - salinity (take a water sample)
   - temperature: air and water
2. Collect fish
3. As each fish is caught, record the following:
   - type of bait used
   - time caught
   - mass of fish
   - total length
   - mass of stomach
   - sketch of teeth
4. Slit the belly of the fish and remove the stomach.

5. Tie a piece of sewing thread around each end of the stomach.
6. Inject the stomach with preservative.
7. Either place the prepared stomach in a labeled jar or attach a label to the stomach and place in a group container.
8. Repeat step 1 - 7 for each fish caught.

B. Later back in the lab.

1. Remove the stomach from its container and place it in a petri dish.
2. Slit the stomach and empty contents into petri dish. Rinse and decant.
3. Count and identify items present.
4. Remove a portion of the loose material trapped in the folds of the stomach wall, prepare a wet mount, and observe under the microscope.
5. Count and identify organisms.

GRAPHIC ANALYSIS

1. Graph the mass of a fish against its stomach mass (calculate the average ratio).
2. Calculate the frequency of occurence for each organism identified (number per animal).

QUESTIONS FOR CONSIDERATION

1. What happened to the bait used to capture each fish?
2. What conclusions can be made concerning the feeding habits of species examined?
3. What organisms were most abundant and least abundant in the stomach of each observed species?
4. Describe any observed relationships between type of teeth and food consumed in the analyzed specimens.

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PREPARATION OF HERBARIUM MOUNTS

TO THE TEACHER

In collecting algae in the field, all specimens from a particular habitat should be placed in a plastic bag and numbered to match a data number from the field notebook.

This exercise should begin with mounting and pressing at the end of the week and drying over the weekend in dry chamber or oven. Cards can be dried in an incubator for the weekend. Classification can begin on Monday and continue for as long as necessary or desired. Mounts can be prepared individually or as a composite.

This lab may be run concurrently with the analysis of floating seaweed populations if desired.

These techniques are applicable to fresh water algae and assorted water plants as well as marine algae.

It is suggested that the teacher choose the best specimens each year and use these to build up a school collection.

The teacher may desire to use standard herbarium paper or some other cardboard stock rather than file cards in order to secure larger specimen mounts.

An Alternative: Xerox Method

A disadvantage of herbarium specimens comes in two forms: insects and kids. Insects eat them and kids, with repetitive handling, tear them up. With some experimenting with darkness setting on a photocopy machine, you can obtain excellent reproductions of marine algae and plants. These algae photocopies will have a much longer lifespan than the standard herbarium mounts and can be very useful in the classroom. Float the delicate specimens onto a plain white sheet of paper before photocopying. An intermediate step of slow-drying the specimens in a plant press for a few days, allowing them to dry, will remove frustration on your part.

TO THE STUDENT

As a coming source of food for mankind, algae cannot be overlooked. At present time seaweeds, giant kelp, are being collected and converted to flour. The Japanese are raising *Chlorella* to be used as a protein, fat, and vitamin supplement for their diets.

Apart from its use for the culture of micro-organisms, the extracted agar from seaweeds has a variety of uses. Among these can be included the canning of fish, the sizing of fabric, in paper and glue manufacture, to add gloss and stiffness to leather, in cosmetics and medicines, and as a thickening agent in ice creams, sherbets and pastries.

PURPOSE

To prepare herbarium mounts of marine algae.

MATERIALS

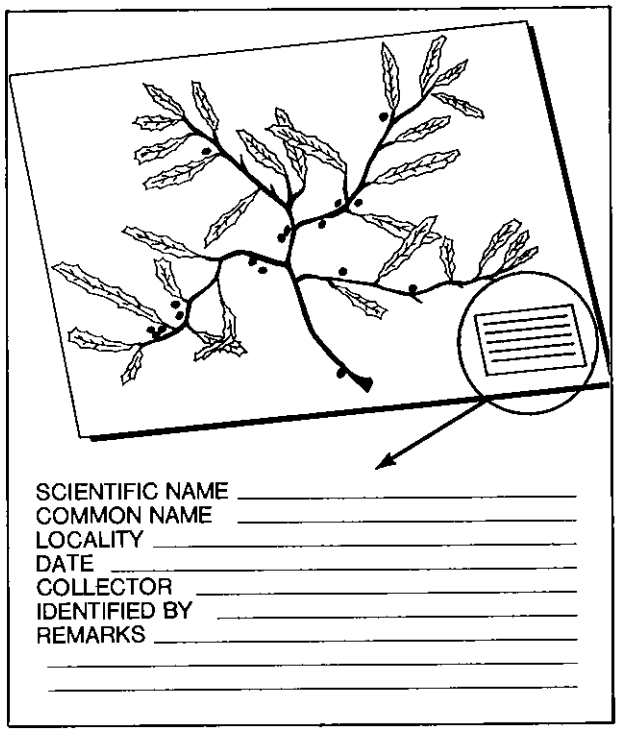
Plant press
Plastic bags and ties
Tags (white paper)
Shallow pans or trays
Probes
Scissors
5 x 8 file cards or herbarium sheets
Wax paper
Cellophane wrap
Newspaper
Drying box
Assorted algae

PROCEDURE

1. Remove a specimen from a numbered bag and record this number on a 5 x 8 unlined file card.
2. To a shallow, flat pan add water to a depth of ¼ inch.
3. Place the algae in the pan and tease it into flat position.
4. Select a representative portion to use as a mount.
5. Slip the card under the specimen and arrange the specimen into desired position.
6. Remove the mount by grasping a corner and raising gently to drain the water. A second method is to mount the paper on a floating board and place in water for mounting.
7. Place the mounted algae into a plant press using alternate layers of newspapers, the algae mount, wax paper, newspaper. Corrugated cardboard should also be used in-between newspapers, to allow air to help dry the mount.
8. Place the press in a drying box or a hot dry place.
9. When the mount is dry, cover it with a sheet of cellophane wrap slightly larger than the mount and overlap the self-adhering portion on the back of the card. (White glue, such as Elmer's, can be thinned to a brushable consistency with water and brushed on the dry mount to preserve.)
11. Mount the preparation on a sheet of 8½ x 11" stock and record collecting data.

12. Secure dichotomous keys and/or refer to the school collection for identification. Record all information as shown below.

Organized Data Recording



QUESTIONS FOR CONSIDERATION

1. What is the educational value of a herbarium collection?
2. How are or can algae be used as indicators for the animal populations or mineral deposits?
3. Of what value is the information recorded on the prepared algae mount?
4. What future benefits to man may be derived from the culture of algae?

LIMITATIONS AND SOURCES OF ERROR

1. Algae need more pressure applied than standard botanical mounts.
2. If the newspaper and waxpaper are not changed on a regular basis, the algae will begin to adhere to the waxpaper.
3. Extreme care should be used in selecting the portion of algae for mounting.
4. Wasted time if not properly scheduled.

RESOURCE MATERIALS

Algae will be found almost universally distributed through the shallow, off-shore areas, on reefs, around mangrove hammocks, bays and estuaries.

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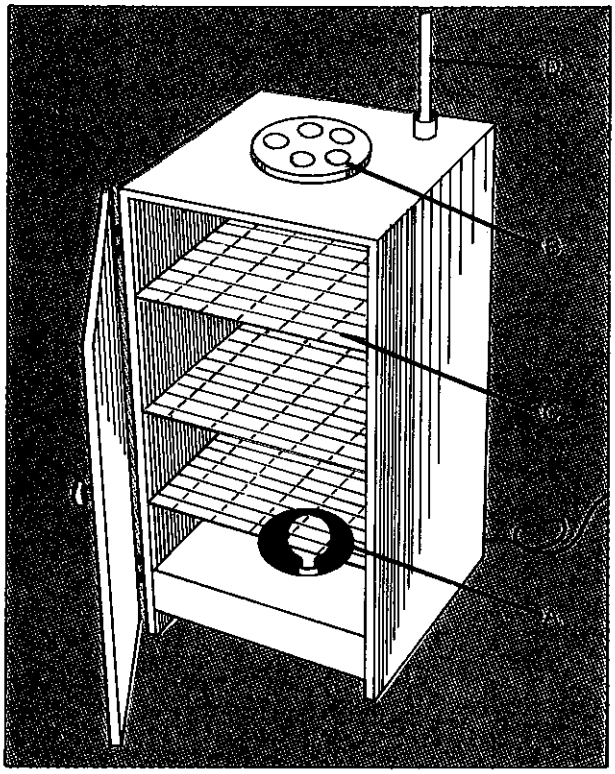
HERBARIUM AND INSTRUMENT DRYING BOX



A rectangular box of ½" plywood built in size to accommodate one or more herbarium presses. The size is dependent upon available space and need.

Item A in the illustration represents a 40 to 75 watt light bulb which provides heat for drying.

Item B is a series of air holes for moisture-laden air to escape. These should be drilled to some available stopper size as a No. 5, 6, or 7, or wheel-shaped closure as illustrated. Item C represents a wire shelf. This should be placed a little below the center of the box, so that items placed on it will be in the approximate center. If desired, more shelves can be added. Discarded refrigerator racks can be used as shelves. Item D represents a thermometer.

The box illustrated here is 28" w x 28" d x 38"h. The closure wheel is 10" in diameter.

The drying box is also effective for drying instruments. If the instruments are rinsed in running water after use with salty marine specimens, much of the rusting usually associated with these operations can be avoided.

BEHAVIOR OF MARINE ORGANISMS

INTRODUCTION

An old story starts with a character looking up the word "Ecology." He discovers that his dictionary says "the study of cycles." Perplexed, he looks up "cycles." "To go in a path so that you end up where you started", he reads. He then goes back and looks up the word "Ecology."

Ecology might better be defined as the interrelationship between organisms and their environment. The interactions between organism and organism, between organism and physical environment is what will be studied in this unit. The emphasis will be on behavior. No ecosystem is better suited to studying the wide spectrum of interactions than the sea.

A function of behavior exhibited by marine organisms is the territorial imperative. Many organisms stake out a territory and defend it against any intruder. Some organisms modify their territory considerably to suit their living needs.

Non-living stimuli from marine organisms' surroundings can also cause behavioral responses. Grouping is a common behavior in response to such factors as light, wind, food, current, temperature, strange movements and objects, moon phases and tides. Sometimes groupings have an economic importance to man. For example, a coral reef can sink man's ships or attract fish which brings him food. In this unit behavioral responses to different environmental stimuli will be studied.

TO THE TEACHER

Look for local people for help on which local animals are most easily obtained and most suitable for those procedures. Try pet stores, scuba divers, county marine extension agents, science coordinators, environmental groups, fishermen.

Always be prepared to utilize aquarium disasters. If something is eaten you have an excellent example of predation. If two fish fight over the same food, you have competition. If a fish is infected by parasites, you can study parasitism.

If you are fortunate enough to take field trips to the sea, take time to do behavioral studies. Many of the procedures in this unit could be modified for field trips. The behavioral activities listed below should be of high interest for students and lend themselves as springboards to many concepts. Get your feet wet and expand on these ideas.

1. How does changing aquarium temperature affect the behavior of fish?
2. How does a fish react to colors?
3. How do different fish express aggression?
4. Why do brittle stars hide under rocks?
5. What organisms group toward light and which ones group away from light?
6. Have a fishing lure contest and then a fishing tournament to see how they work.
7. Have a group of students use the same lure but each retrieve it in different ways.
8. Keep small schooling fishes in a large aquarium and study behavior of schools.
9. Does a crab behave differently when he's missing a claw? (Keep crab around awhile to show regeneration.)

TO THE STUDENT

Many types of interactions involving organisms can be studied quite well in aquaria. Below are some specific suggestions.

A group of fish common to Florida reefs and cheap in pet stores are damsel fish. These are good fish to use in procedures involving territoriality, especially those in the genus *Pomacentrus.* Try using the beau gregory *P. leucostictus,* the threespot damsel fish *P. planifrons* or the sergeant-major *Abudefduf saxatilis.*

PURPOSE

1. Study territoriality in fishes.
2. Classify fish reaction to objects dangled before it as "Look-Bite-Ignore."

MATERIALS

20 gal. (or more) aquarium with plenty of hiding places and grid laid out on surface of sand
Damsel fishes, assorted
Assorted small objects (see procedure B)

PROCEDURE

A. Territories
   1. stabilize aquarium
   2. put in one *Pomacentrus sp.* fish
   3. allow one week for fish to acclimate
   4. drop small object into each grid space in turn, allow fish to settle after each drop
   5. diagram aquarium grid and place an "x" in each space where fish attacked dropped object

6. try different objects and repeat steps 5 & 6
7. allow fish 24 hrs. minimum to settle
8.introduce a second fish of the same species and compare size to the aquarium
9. map the territory of each fish immediately after introduction
10. map the territory of each fish 3 days after introduction
11. remove second fish
12. allow fish 24 hrs. to settle
13. introduce a different species of fish, comparable size, preferable *Abudefduf* sp. and repeat steps 9 & 10
14. you may expand this experiment with additional fish
15. construct a stone wall up aquarium side. (remove damsel fish before doing this)
16. lay out grid on aquarium side opposite wall
17. repeat territory experiment using blennies and vertical territory

B. Look, Bite, Ignore
1. stabilize aquarium
2. put in fish (try different species)
3. allow one week for fish to acclimate
4. develop grid-type data sheet with fish names down side, object names across top
5. dangle object in front of each fish in turn allowing time for fright reaction to subside
6. record response to object by each fish in appropriate space on a data sheet as LOOK, BITE or IGNORE
7. repeat experiment with different species and with territorial versus free-ranging fish
8. repeat experiment using crabs instead of fish
9.develop a fishing lure for one fish species using their response to objects to maximize lure appeal

PURPOSE

Study behavioral responses of echinoderms to external stimuli.

MATERIALS

20 gal. (or more) aquarium set-up, assorted sea urchins, sand dollars, starfish

PROCEDURE

A. Echinoderm Races
1. turn several sea urchins on their backs and time how long each takes to right itself
2. use different species in the same race
3. use starfish of same and different species in a race
4. place food at one end of aquarium and time how long it takes urchins or starfish to reach it (don't race urchin and starfish in same aquarium)
5. place sand dollars on surface of sand and time burrowing speed

B. Sea Urchin "Masking"
1. place grid in aquarium
2. place small objects commonly found on sea floor, (shells, sticks, leaves, pop tops) in aquarium, one to each grid space
3. make grid type data sheet with urchin names down side and object names across top
4. place urchin in the middle of the grid
5. document reaction to objects as
G = goes around it
W = walks over it
K = keeps it
P = passes it over back
6. repeat with different objects
7. repeat with objects in different positions
8. repeat with different urchin species

QUESTIONS FOR CONSIDERATION

1. How do the territory sizes compare between: a) new and old fishes; b) fishes of different species; c) among several fishes introduced at the same time?
2. Would the size of the territory grow as the fish grows?
3. Was there difference in response to dangled objects by different species of fishes.
4. Why do you suppose different fishes prefer certain objects (BITE)?
5. Did fishes attack objects that dropped gently through the water more often than objects that plummeted to the bottom? Why?
6. Why was there a different response to dangled objects by territorial versus free ranging fishes?
7. What anatomical and physiological features allow urchins to turn over? Starfish?
8. Why not race starfishes and urchins in the same aquarium?

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ADAPTATION AND FUNCTION

TO THE TEACHER

Correlating anatomy, physiology and behavior is an important and useful part of the concept of ecological niches. Having students examine different members of the same class of animals and relating adaptations to function will help to place the animal into the niche it fills. In this particular lab we have selected the Crustacea for study.

TO THE STUDENT

The decapod (deca = ten, pod = foot) crustaceans are perhaps the most important animals in the detritus-based food web of estuaries, reefs and beaches. They are the middlemen, converting plant matter which most other consumers cannot digest into animal protein in a delicious package. Most consumers, including humans, enjoy eating crabs, shrimps, crayfish, lobsters and other decapods.

Using the same principles in this exercise, one can generate further study and projects. Reef fishes, for instance, have gone through the same adaptive process.

This activity will first require collecting a variety of decapods, then observing them to determine how their body shape and structure "fits" their functions.

MATERIALS

Several varieties of decapods
Plastic buckets
Magnifying glasses
Small nets on handles (like aquarium dip nets)
Seine net
Tennis shoes
Field book for notes

PROCEDURE

A. In the Field:

1. Locate and capture two or three specimens of each type and immediately examine their physical characteristics. Consider:

Body shape —
- squared or rounded?
- boxy or long, narrow?
- thick or thin?
- pointed or blunt?
- color?

Main (food-getting) claws —
- large or small in relation to rest of body?
- weak or strong?
- long or short?
- are both the same size?
- are both the same shape?
- sharp edges or blunt?

Other claws —
- shaped like paddles (swimmerettes) or pointed?
- hairy or smooth?
- strong or weak?
- long or short?

Collect as many different kinds of decapods as possible. Good examples include:

blue crabs	*Callinectes, Portunus*
fiddler crabs	*Uca*
dock crabs	*Aratus, Sesarma*
land crabs	*Cardisoma, Gecarcinus*
ghost crabs	*Ocypode*
rock or stone crabs	*Menippe*
pea crabs	*Pinnotheres*
arrow crabs	*Sternorhynchus seticornis*
spider crabs	*Libinia*
swimming shrimp	*Penaeus*
snapping shrimp	*Alpheus*
grass shrimp	*Panulirus argus*
hermit crabs	*Pagurus*
sand flea	*Emerita talpoida*
banded coral shrimp	*Stenopus hispidus*
mantis shrimp	*Squilla empusa*

2. Re-examine the area where the crustacean was found. Try to use the most descriptive short sentences describing the immediate surroundings, Example:
   a. on reef, plant, mangrove root, in mud, in sand, in grass flat, etc.
   b. in or out of water
   c. method of capture
   d. available food in the area
   e. and so on

3. Unless there is a strong reason for keeping the animal, quickly replace it where it was found.

So far, you have been concerned with catching decapods and making some accurate observations about them. Now we will try some scientific guesswork (deductions) based on our observations to predict what your decapod eats and how it gets its food. Refer to the data to make intelligent guesses; for example, a crab which has strong claws and swimmerettes is well adapted to catch small fish but not very well adapted to crushing oyster shells. A crab with hairy, skinny legs which

end in sharp pointed claws is well adapted to running, climbing and digging on land but not to catching little fishes. Complete the table below:

DECAPODS OF _____ (locality) Date ____

Name	Body Shape	Main Claws	Other Claws	Habitat

B. Back in the class:

1. Examine your field notes and fill in missing information.
2. For each decapod write a short paragraph describing the following functions:
   - locomotion
   - defense
   - relationship with others of the same species (solitary animal or schooling animal)
   - hole dweller, free swimmer or occasional burrower
   - what does it eat and what eats it
   - reproduction

QUESTIONS FOR CONSIDERATION

1. Realizing that your collecting trip barely touches the surface of this class of animals, guess how crustaceans have to adapt to survive on:
   - rocky coasts with a crashing surf (consider the spindly arrow crab); wide mud flats that dry out for hours at low tide; deep cold nutrient rich waters (Alaska); in the Sargassum mats in the Gulfstream?
2. Realizing that crustaceans are favorite foods for other animals, how would a fat and slow crab, a soft-bodied shrimp type, or a pea crab (living inside an oyster) survive?
3. If you noticed the colors of the collected animals, relate them to food-getting and defense.

ZONATION IN MARINE ENVIRONMENTS

TO THE TEACHER

The daily rise and fall of the tides has created two types of zonation easily visible to your students: vertical, as along a bulkhead or on a piling, and horizontal, as on a beach. Both are easily investigated with minimal materials and are easy subjects for numerical, graphical and pictorial data analysis. This unit can be a part of almost every field trip your students take.

TO THE STUDENT

Zonation refers to the habit of organisms to live in "bands", generally according to how wet that band is. For example, an organism living close to the normal low tide level will be wet a lot more of each day than one living close to the high tide level.

Investigations of zonation are usually done along a line known as "transect", which crosses through all the zones. We will describe the use of a transect for both vertical and horizontal zonation.

PURPOSE

To observe, describe and analyze zonation in marine environments, and apply other ecological concepts such as adaptability, density, diversity and limiting factors to explain zonation.

MATERIALS

Heavy cord, cut to suitable lengths for areas to be studied
Clipboards or notebooks, paper and pencils
Magnifying glasses
Face mask or glass-bottomed bucket
Metric rules
Field guides
Camera
Tennis Shoes

PROCEDURE - PART I - *Vertical Zonation*

A. Select a vertical surface, such as a rock outcropping, piling or bulkhead. Try to work at low tide.

B. Lay a transect line with your string, starting below the low tide and going straight up above the high tide line. Generally, half a meter above and below the tide marks will work well.

C. Identify the more common (dominant) organisms along your transect line. You should find at least two or three in each zone.

D. Determine a reasonable sample interval for your transect line. If the tide rises and falls only about .5 m, a 10 cm interval might be suitable, but if your area's tidal range is 1.5m, a 20 or 30 cm interval might work better.

E. Make a scale drawing of the transect line. A scale of 10cm = 1m should work.

F. Starting at the bottom, record the organisms present within 5cm on either side of the line at each sampling point. Do this by laying a ruler perpendicular to the line and recording those organisms under the edge of the ruler.

G. Indicate Mean High Water (MHW) and Mean Low Water (MLW) on your scale drawing.

PART II - *Horizontal Zonation*

A. Try to do this at low tide.

B. Lay a transect line with your string. It should stretch from the edge of the water (or the low tide level) into the vegetation above the high tide line. Your investigation may also include zonation here.

C. Determine a reasonable sample interval for your transect line (preferably not less than 10 intervals).

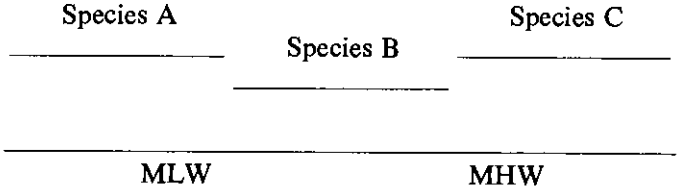
D. Make a scale drawing of the transect line.

E. Record the organisms present at each sample point. Local conditions will determine how large an area or how long a line should be used at each point.

F. Identify and record organisms at each sample point.

GRAPHIC ANALYSIS

A. Develop a chart of "indicator organisms" for each zone by showing the sample points at which each species was found, as below:



B. Develop a Diversity Index to show change in species numbers from zone to zone. To compute this value, record an "x" for each specimen of the same species, then an "o" for each specimen when the species changes, and repeat.

Example: Four of one species within a zone followed by six of a different species in the same zone and 3 of a third species would be noted as follows:

XXXX OOOOOO XXX

$$\frac{N}{a^2 + b^2 + c^2 + \ldots}$$

N =Total Species
a^2 =Species 1
b^2 =Species 2
c^2 =Species 3, etc.

$$\frac{13}{4^2+6^2+3^2+} = \frac{13}{16+36+9} = \frac{13}{61} = .21$$

The closer the diversity index is to 1, the greater the diversity. A seashore biological community of great diversity is generally regarded as more stable and "healthier". Communities of single species would also give an index of 1 and indicate a mono-culture. Disease, predation and environmental stress could easily destroy a community structure of this type, making it far more fragile.

QUESTIONS FOR CONSIDERATION

1. Which organism is predominant in each zone?
2. What feeding adaptation has each organism made to function in its zone?
3. Has the organism made special adaptations to attach to the surface? Describe.
4. Which class of algae lives closest to MLW? to MHW?
5. What factors limit life in each zone?
6. Which zone has the highest Diversity Index? Why?
7. Which zone has the highest overall density of organisms? Why?
8. Can you find evidence in the distribution of organisms of the effect of spring and neap tides? Describe.
9. If possible, compare zonation on man-made objects with zonation on natural surfaces. Especially compare densities and Diversity Indexes.
10. What is meant by supralittoral, infralittoral, sublittoral, eulittoral?

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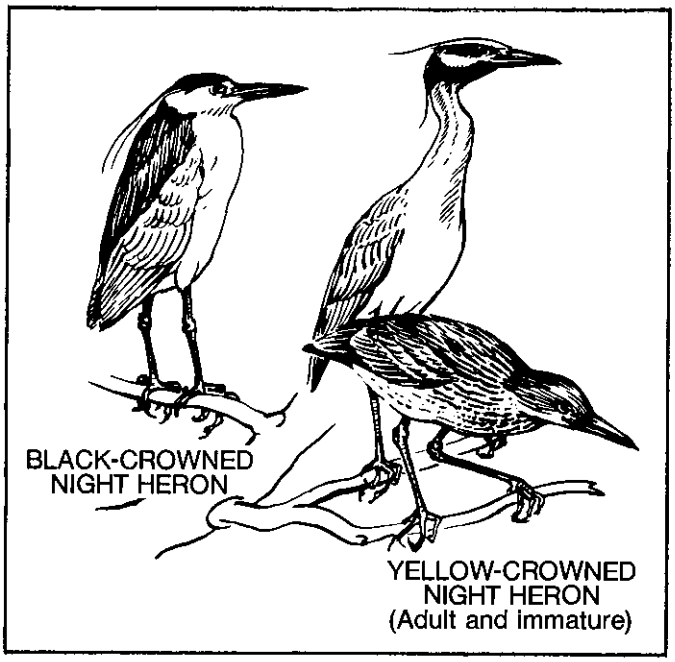
STUDYING COASTAL BIRDS

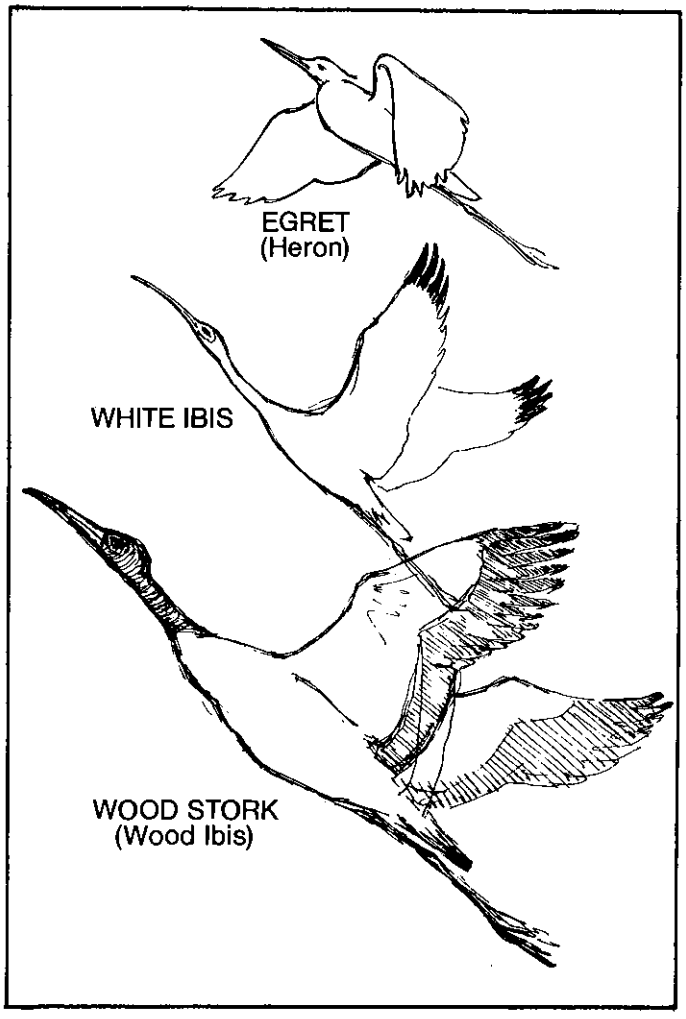
TO THE TEACHER

Bird study is a pushover. Our "feathered friends" are such beautiful and intriguing sentimental favorites of the people that thousands of groups are organized across the country for their admiration.

This should all be to the advantage of a unit on bird study but actually presents problems. Most of the effort of organized bird study is guided by a nationwide "bird golf" psychology: keeping score on species seen. The game becomes competitive as epitomized by the annual "Christmas count." The fevor of competition is so strong that scientific accuracy can be severely stressed by the emotional desire to build an admirable "score" of species.

No one wants to interfere with this pleasant and beautiful outdoor recreation. It has much scientific value as well, for so much manpower is devoted to it that a tremendous mass of data is accumulated . . . far more than scientists could ever collect otherwise. We know much about which birds are where . . . and when.





However, a teacher should quickly get beyond this concept, toward answering "why" some bird events happen. Put your students in touch with the local club. Meanwhile move into studies which approach these basic truths:

1. Birds are integral parts of the ecosystem, therefore of food webs, just as surely as earthworms and other less spectacular organisms.

2. Their biomass is fixed by food availability and the series of other limiting factors that control each species.

3. By differences in their natural equipment and in their behavioral patterns, bird species are adapted to unique niches which often vary seasonally.

While these principles sound pretty abstract and uninspiring, their proof in the field is exciting.

THE FAMILY SPECTRUM PHENOMENON

Within a family grouping there will often be a series of bird species built on the same body plan although in a sequence of sizes. Herons are a dramatic and easily observed example of

this concept. There are likely to be half a dozen common species in an area, ranging from the Green Heron to the Great Blue. Knowing the leg length of each species an observer could estimate the depth of water in which a bird is fishing and perhaps the size of a fish caught before it disappears down the throat. Notes on comparative hunting techniques could be kept.

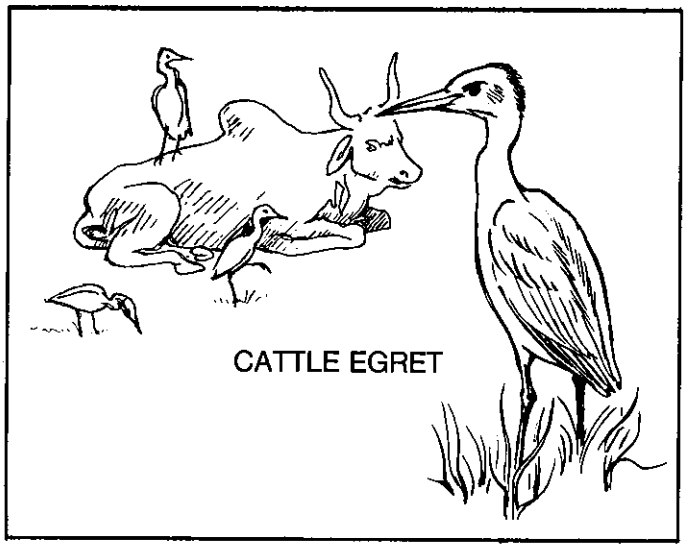
In other words, the whole group, taken as a team of predators working over a variable prey population, makes a challenging adventure. The unique long vertebral bone in the neck turns out to be a valuable part of the successful fishing technique of these spearfishermen on stilts. They'll learn, however, that the fish are not speared but caught between the mandibles, and not swallowed willy-nilly but always *headfirst*, for practical reasons.

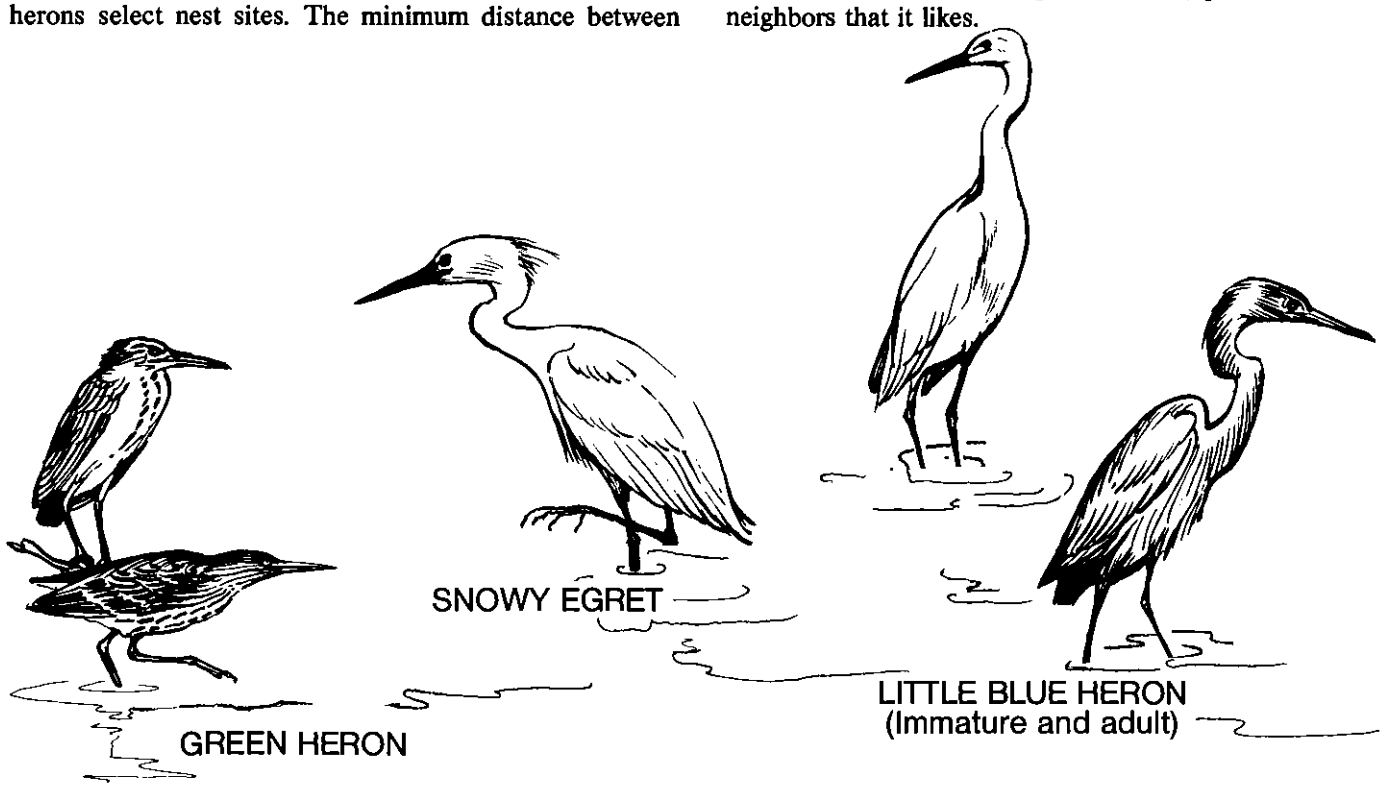
Terns make another good study of this type for there are often Least Common (or other medium-sized) and Royal Terns around. With binoculars one can often see the fish just caught on a plunge. The fishermen call these birds "strikers" because of their fishing style, which might be compared to that of the Brown Pelican and the Osprey as to percent success per dive, prey size and frequency of attempts.

In fact, if a class got ambitious, a whole mosaic pattern could be worked out to show the overlapping but separate geographic and prey-type niches of all the fish-eaters in an area in a given season.

The two groups mentioned also show dramatic nesting colony behavior which lends itself to documentation and analysis. The big question here is whether the birds can be observed without disturbing their breeding success. If so, it can be shown that a "caste system" exists in vertical stratification and in mixing of species in the same tree as the herons select nest sites. The minimum distance between nests is the thrusting distance of the saber-like bills of the nestlings. If the off-beat Cattle Egret has been overlooked in the studies of other herons (fish-eaters), the rookery brings it back together with its relatives, and as a recently arrived immigrant it shows no bashfulness in taking its place in the colonies.

The terns likewise show a distinct zonation of species as they scoop out hollows in the sand for their eggs in closely-clustered colonies. As plant succession alters the area, the colony will shift so that each species continues to have the specific characteristics of ground cover, particle size and neighbors that it likes.



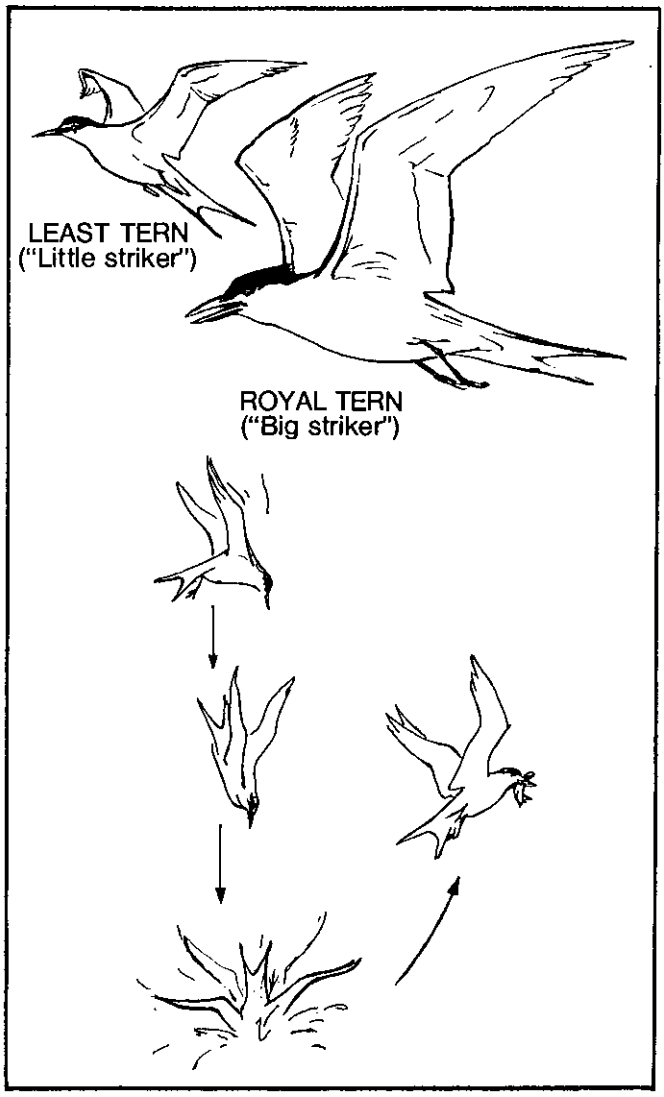


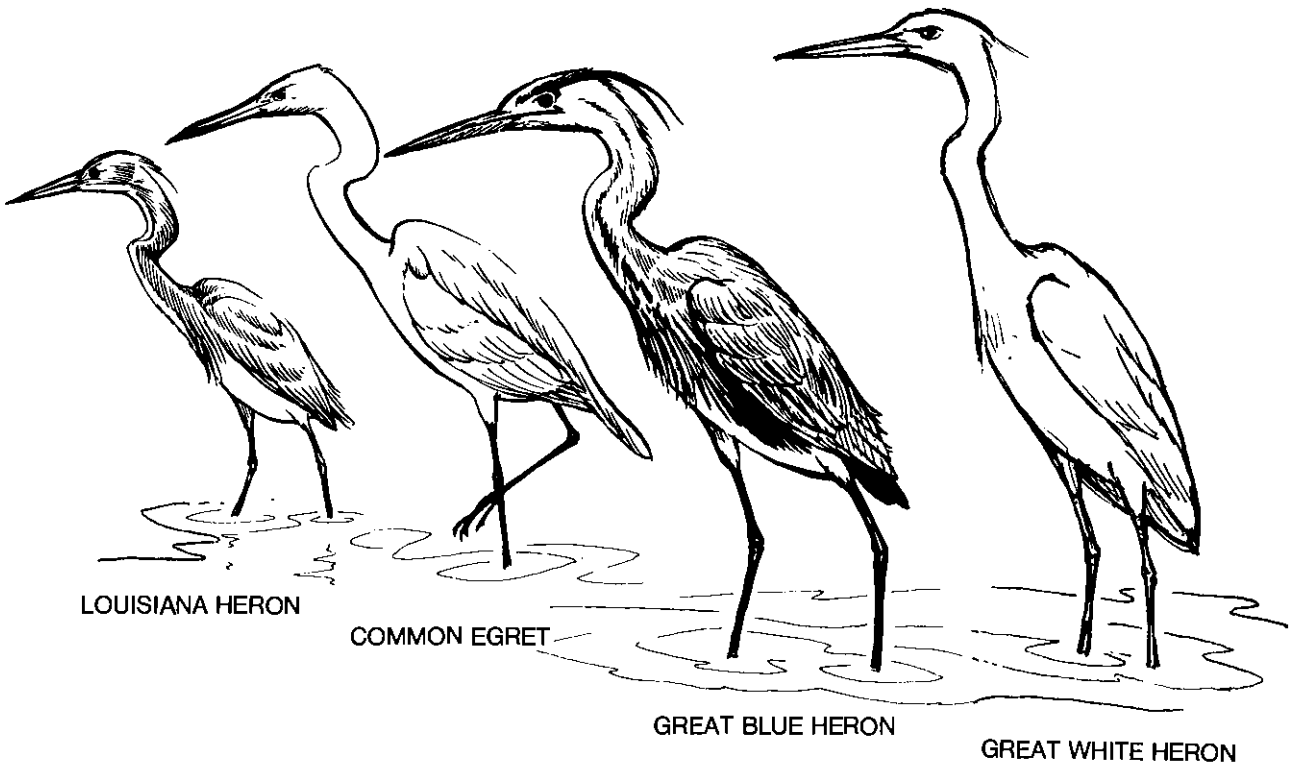
In all of these studies a year-to-year follow-up using uniform technique would provide data suitable for publication. Little of any of this is in the literature and a long-term study unheard of. It is the type of routine observation requiring much manpower and not very attractive to funding agencies. There are many much simpler studies for a couple of weekends of time invested.

SOCIALIZATION

All birds flock at times, although in watching Great White Herons bicker you wonder how any two ever occupy the same acre of ground. Continuation of the species proves that they must at times tolerate each other. Some species ordinarily work in a solitary manner, roost together while loafing, flock up seasonally when moving and especially when nesting. Herons and Terns and Gulls more or less fit this pattern. Others, like Sandpipers are almost opposite, breeding individually and clustering to feed. One of the most fascinating but easy studies is to analyze birds for congeniality. Some factors which may be specifically picked apart as distinct aspects of behavior are:

1. Number of birds of same species in the flock.
2. Relationship to other species, which may even be part of the flock.
3. Interplay between individuals within the flock.
4. Group dynamics; that is, are they moving in a group (following a leader?) or just casually associating?
5. Daily rhythms of social behavior and seasonal shifts if the project is long-term.
6. Flock response to interference (by observer, for example).

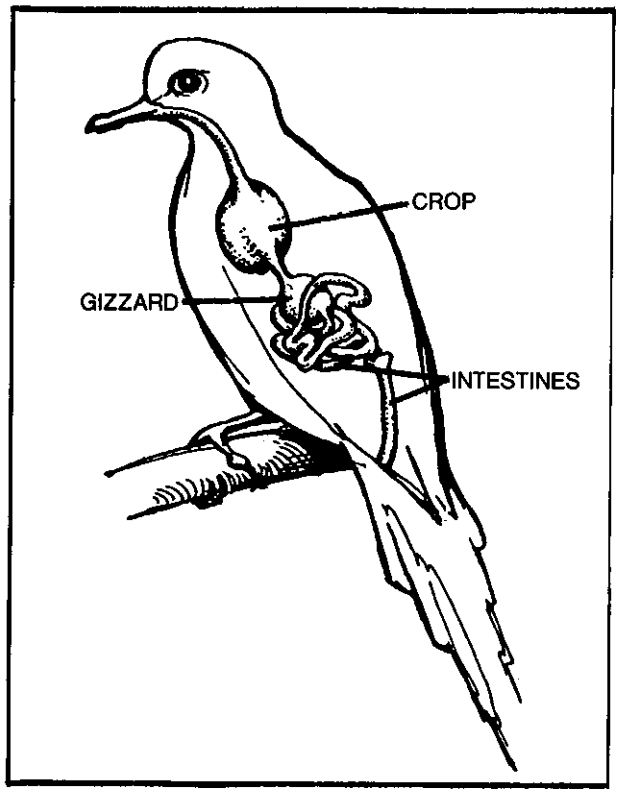




FRIGHT: THE FLIGHT RESPONSE

Given just an afternoon or two an observer can complete an interesting contrast between bird species in their attitude toward human beings. To show how tricky this is, remember that the ducks feeding at your feet by Daytona Beach seawall or at a Winter Haven lake would not allow anyone within a hundred yards of them anywhere else a few months later. Geese, in fact, will fly over a hundred yards high over areas surrounding a small refuge pond and then drop down sharply as they pass the boundary line and the hunters. So we know that the instinctive behavior can be modified by learning in specific situations.

In its simplest form, however, this study seeks the normal inherent flight distance of each species. The student may discover some strange selection in the process. For example many species which are fidgety in the presence of an approaching person on foot will let a car get much closer, apparently not computing it as recognizable danger. Likewise vultures sometimes get conditioned, in feeding on road-kill animals, to holding their ground as cars whizz by 20 feet away. However, when a car stops this is not part of their conditioned tolerance, so they flush.



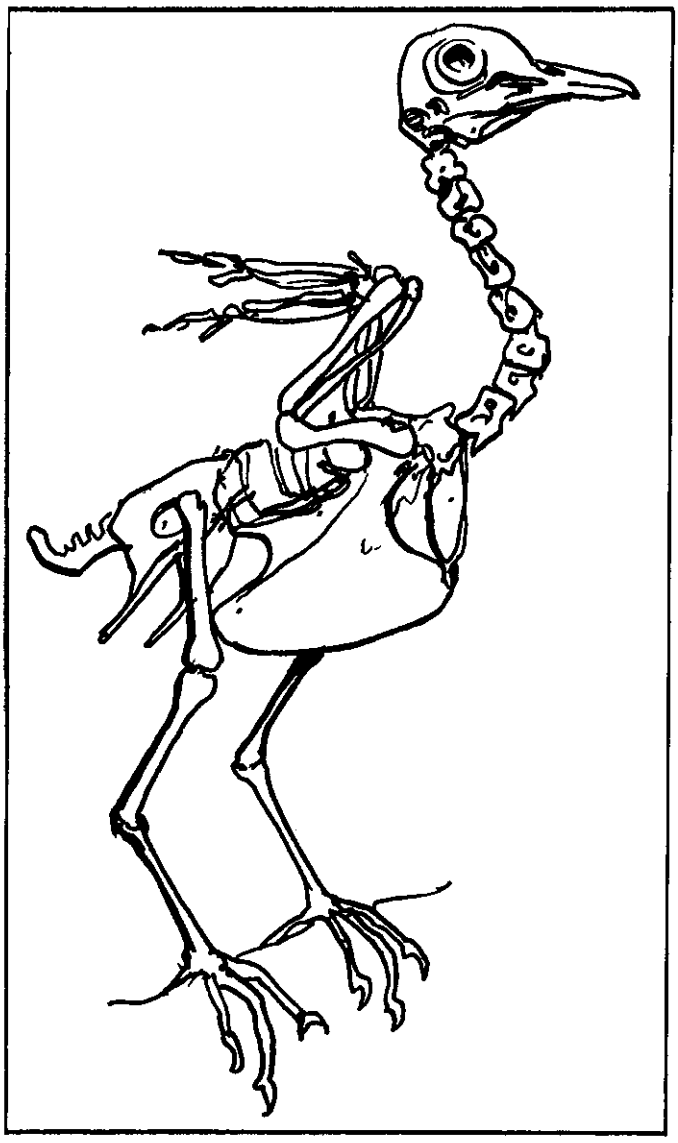
FOOD HABIT STUDIES

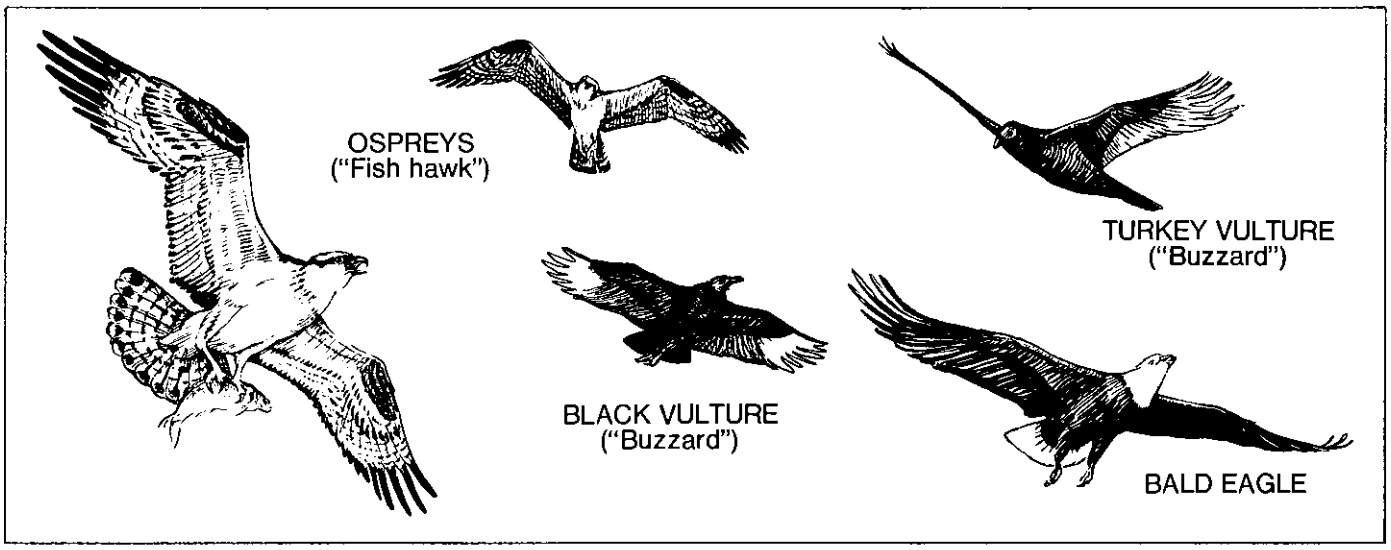
More stomach analysis needs to be done, to follow up the now ancient studies of Martin and Zim, but your students shouldn't do them unless there is a ready source of fresh specimens. Collecting should be discouraged for all projects, but there are TV tower kills and other accidents from which specimens can be obtained. Legal hunters are a good source for game species. These are, however, the species on which data is already most available.

Likewise bird-banding should be discouraged, for it is a carefully-regulated and legally-restrained activity. Someone in the Audubon Society with a master permit may be willing to help a teacher or a very serious student with long-range objectives, but look for other methods first. Learn from and work through the established banders if you decide the technique is needed.

Gut analysis is tedious and difficult. By the time food has passed a bird's gizzard it is largely bent out of shape, although seeds can often be identified if one has a reference collection of seeds from known plants. That requires a heap of doing.

Another project calls for a reference collection of bones, for hawks and owls conveniently cough up their stomach contents for you. These pellets are found beneath the nests or feeding perches and are ready-made projects. Identification of the bones and fur can be approximate, the study being of numbers, size and variety of prey. In the case of the Osprey the pellets will contain fish bones and scales, of course.





THE MORE ROUTINE STUDIES

So far we have largely skirted the obvious, which is to measure and record either anatomy or mannerisms of bird species. These things all lead to improved identification, are helpful in developing skill at observing and are easily designed.

Comparative study of bills and/or legs in a museum set of study skins (or in live birds) is a natural. A detailed study of feathering is very illuminating when related to functions of the various types. Along these lines, a study of molting would make a good study for a serious student with some months to watch. For example, Laughing Gulls change dramatically as they go through either spring or fall molt, and the immatures will add another dimension since they have distinctive plumages.

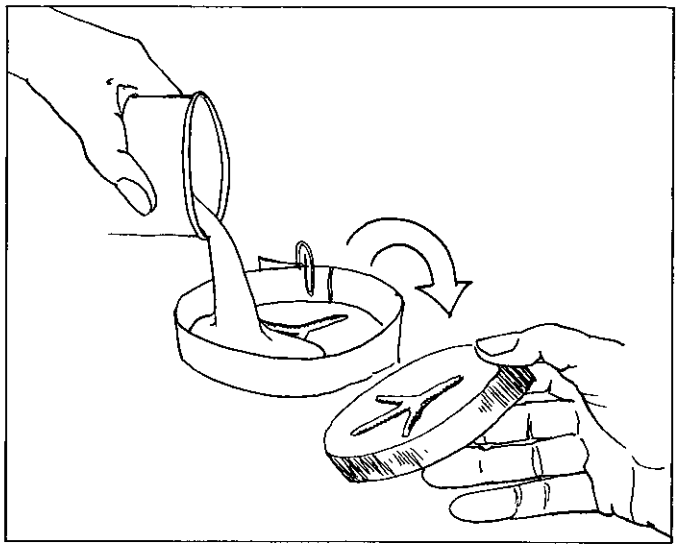
Flight patterns make simple but interesting studies for someone able to sketch what is observed, and the songs of birds would be a natural for someone who knows music well.

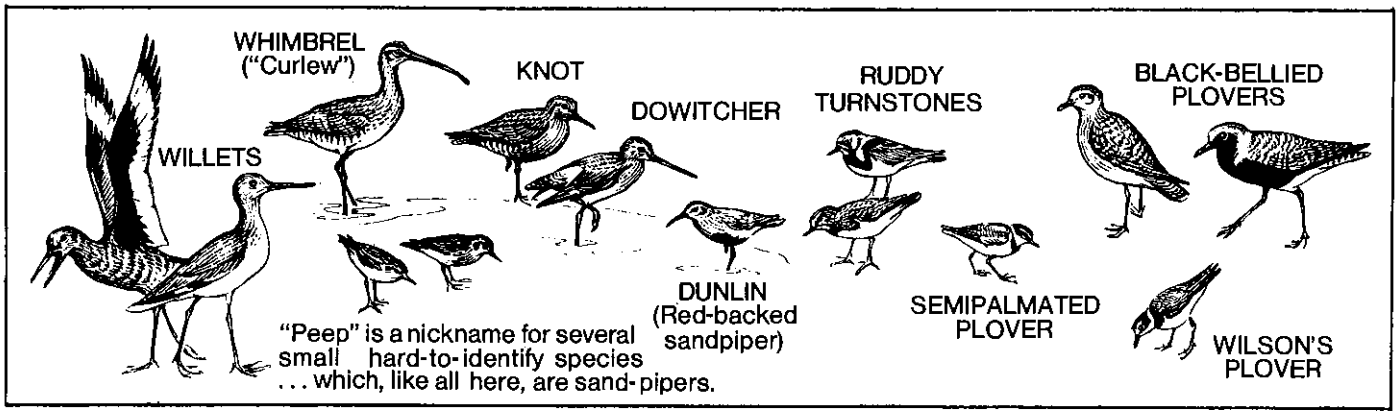
AND THE HANDICRAFT PROJECTS

Bird houses are too well documented to need much discussion here, except to point out that in Florida most are probably occupied by Starlings or English Sparrows if by anything. However, well-constructed Bluebird houses and Wood Duck boxes can be very useful, so get instructions from the experts on exact dimensions.

Casting plaster molds of bird tracks is still fun and teaches a generally useful technique. Fiberglass and other modern materials may be better for your purposes. Since it takes students out and requires a tangable product on return, such a project has real merit for some young folks.

Keep away from nests and eggs, of course. Even poking





around nest sites and leaving them "unharmed" can be disastrous for the brood, due either to upset parents or a predator following your scent. Observing nests from a distance and collecting nests out of season can both be good projects, but in actual practice, often lead to saddening experiences.

If a student is inexorably pre-med and must operate, don't be discouraged at the absence of a victim. Have him buy a chicken at the grocery store, boil it for two hours and have him pick the bones clean. Let them dry thoroughly. And all that is the easy part. Now with airplane glue reconstruct the skeleton (you'll need a couple of props to help stand it up). Some of the bones can be sawed in half to show the incredibly delicate and lightweight construction.

SUMMARY

All told, the most fun for you and students will come when you push them outdoors and lead them toward ecological studies. Problem one: they'll have a hard time pinning down the parameters that define their study and therefore become the data. Problem two: with a hint of frustration beneath their pride, they'll always say that they finished with more questions than they started with, but that's good science.

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NEWS OF MARINE SCIENCE

INTRODUCTION

The newspaper is in fact the greatest assemblage of information compiled and issued for general public consumption each day. In many cities the cost of the daily paper is still a quarter or less. A 50-page newspaper contains the word equivalent of a 250-300 page paperback book. With most paperbacks books costing $2.00 or more, a 50-page newspaper is indeed a real reading bargain. It is a "living text book" printed new and fresh each day. For millions of adult readers it is *the* primary source of new information for updating past instruction and education. It can also serve the same function for the child, the adolescent and the youth while in the classroom.

TO THE TEACHER

A close look at a newspaper published in a coastal city (community) will reveal that many items are of continuing interest and concern to the marine science student. The tide, the water temperature, the sea condition and the marine weather are regularly reported, usually appearing with the weather map and forecast. Events such as tropical storms, hurricanes, waterspouts, etc., get special news treatment even though these may not be local phenomona. Environmental concerns of the community, such as endangered coastal wildlife, commercial fishing news, erosion problems, coastal planning, etc., often appear as feature stories in Sunday issues. Special scientific exploration work, environmental disasters, such as tidal waves (tsunamis), whale beachings and the like, which are often reported by the news wire services, receive front-section coverage in many dailies.

With this wide array of information-resource material as a starting point, the class or each student can pursue items of interest (concern) to whatever depth he/she desires. In some reporting, opinions are given for facts are not precisely stated in an article. This too can be a matter for student research and verification — and a useful learning experience. It can be a catalyst to the individual formulating his/her own opinion based on additional facts and new findings. Environmental news should be of concern to everyone. It is essential to keep up-to-date on improvements as well as deteriorating conditions. Only with up-to-date information can one arrive at a reasonable conclusion and comment knowledgeably regarding an issue. There are any number of exercises that can be developed from this resource. Only one is given here. This type of project is tailored for the serious science student(s) and is intended to advance individual learning and research skills in preparation for later individual study. It is designed to be a semester long activity to be integrated with other classroom requirements.

TO THE STUDENT

Marine science is a conglomeration of many separate sciences: physics, chemistry, biology, ecology, geology, engineering, geography and cartography, and has a very close linkage with certain aspects of the social sciences. In a study of marine science, it is important to note the interrelationships of environmental events and other current events in the world about us. Many news reports serve as "case studies" (applications) of environmental concepts learned in the classroom. In addition, one must learn to probe further into possible solutions of unsolved problems and unanswered questions. Newspaper articles often present an opportunity for the student to undertake further investigation on local issues. This is the challenge and the intent of this exercise. It offers a chance to do research on a current topic of your own choosing.

EXERCISE I

PURPOSE

To acquaint the student with contemporary activities, advancements and concerns in marine sciences and related topics.

MATERIALS

Daily (incl. Sat. & Sun.) subscriptions to the local newspaper and those of several nearby coastal cities. A minimum of 5 copies of each paper is needed. It is suggested that subscriptions be placed with at least 4 different papers (6 copies of each is recommended for a class of 30 students).
Scissors (15-30 pairs depending on class size)
Paste (as required by class size)
Looseleaf scrapbooks (as required by class size)

PROCEDURE

(To the Instructor: This project can be done as individual studies or in team groups. The latter arrangement may be the most beneficial, if teamwork prevails among the students in each unit.)

Divide the class into teams with 3 to 5 students in each. Allow each group to select a general marine science topic (or a closely related one) for their work. Suggested topics are: marine meteorology; endangered marine life; coastal planning; marine geology; coastal ecology; fisheries; marine recreation; disasters; research and education activities; port development; etc. Team topics need to be broad enough to allow each student on the team to select a specific subject within that topic category for his/her further examination.

For example: A team of 3 students who chose marine meteorology, might select the following for their individual projects: Hurricanes; local coastal climate, and unusual marine weather conditions. One class period per week should be devoted to examination and selection of news articles which are relevant to the topic selected by the student. Team cooperation is helpful in reviewing the weekly collection of papers. Special attention should be directed toward the editorial page, cartoons and syndicated features. Often these focus on environmental subjects. Arrange collected clippings in a looseleaf scrapbook in chronological sequence — note date and the newspaper from which taken.

After 6-8 weeks of collection, work should begin on developing a 10 min. verbal report to the class on the subject. Reporting dates near the end of the semester are best. One week may be set aside for reporting with one day devoted to each team's "topic category." Of course collection of newspaper materials needs to continue. Outside investigation is recommended to relate current events to principles, to previous events, to other impacts on the environment, and to social impacts involving people and communities. Each student's project report should summarize the topic selected and provide a conclusion. In some cases interviews with professionals or public officials can be helpful as part of the investigation process.

Each student needs to prepare the collection of clippings in an appropriate manner for submission and append to the collection a 1,500-2,000 word (6-8 typewritten pages) summary of the subject/topic selected and covered. Emphasis should be placed on "a contemporary view/perspective" with the student's view being the most important end product.

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1975, *Using the Newspaper in Physical Science Classes,* Houston Post, Texas, 33p.

1975, *Using the Newspaper in Life Science Classes,* Houston Post, Texas, 34p.

FILM

The Newspaper and Your Quest for Truth — Film Strip w/cassette, Current Affairs, P.O. Box 398, 24 Danbury Rd., Wilton, Conn. 06897.

INTRODUCTION

When we study marine science or any other related field we must consider the weather. In fact, you probably consider the weather whenever you do anything although you may not think of it directly.

Weather and climate are created by 1) *heat from the sun,* 2) *the rotation of the earth,* and 3) *the different absorption and radiation rates of the land and water masses* on the earth. Radiant energy of the sun and the constant rotation of the earth about its axis serve to set up the circulation patterns of the atmosphere. These patterns carry heat and moisture from the equator to the poles across land and sea and over flat lands and rugged mountain terrain. Patterns of winds, rainfall, cloudiness and temperature tend to be similar seasonally at any given location. Weather sequences generally follow the same patterns year after year. These patterns determine the climate for a given location.

Factors responsible for weather are called *Climate Controls.* These are: 1) *latitude,* 2) *continentality* (land) and *water masses* (ocean), 3) *prevailing winds,* 4) *ocean currents,* 5) *elevation* (altitude) and 6) *mountain barriers.*

Latitude: Heat from the sun reaches the earth after a transit of 93 million miles through space. The rays reaching the Equator come from nearly overhead. Nearer the poles, the sun appears lower in the sky and the rays reach the earth at a sharper angle. This results in warm climates in the equatorial regions and cooler climates toward the poles.

The tilt of the axis of the earth seasonally modifies the latitudinal effect. The earth's axis is tilted 23.5° from a line perpendicular to the eliptic orbit. From about September 21 to about March 21, the north end of the axis tilts away from the sun, so the rays are more slanting in the Northern Hemisphere and more directly overhead in the Southern Hemisphere. Therefore, the Arctic is in continuous darkness in December and January and the Antarctic is in continuous daylight. Still the south pole remains cold since the sun in only 23° above the horizon.

Continents & Water Masses: Land and water areas, when heated by the sun, exhibit different properties. Land surfaces generally heat rapidly. There is little or no penetration, therefore, the land cools rapidly at night. Water warms slowly, and because of its fluidity there is a continuous heat transfer to or from subsurface waters. This mixing process allows the oceans to retain much more heat than the land. As a result the surface layer of air over the continents is warmer in summer and cooler in winter than over the ocean. Because warm air rises there is a net inflow of air over the continent in summer and an outflow from the continent in winter. A similar situation occurs almost daily in Florida and other coastal areas during the summer where the land is cooler at night and warmer later in the day than adjacent water, resulting in a *land-sea breeze.* Oceans, seas and other large bodies of water are significant contributors to the weather and climate of an area. They not only supply the moisture to the atmosphere, but also serve as a heat storage and heat transfer system.

Prevailing Winds — The differential heating of the earth by the sun is the main cause of wind systems. The equatorial regions receive considerably more solar heat per unit of area than do the polar regions. Just as with the continentality effect, warm equatorial surface air rises and is replaced by cooler air flowing in from the poles. The unequal heating of the earth's surface together with its rotation produces the general global circulation.

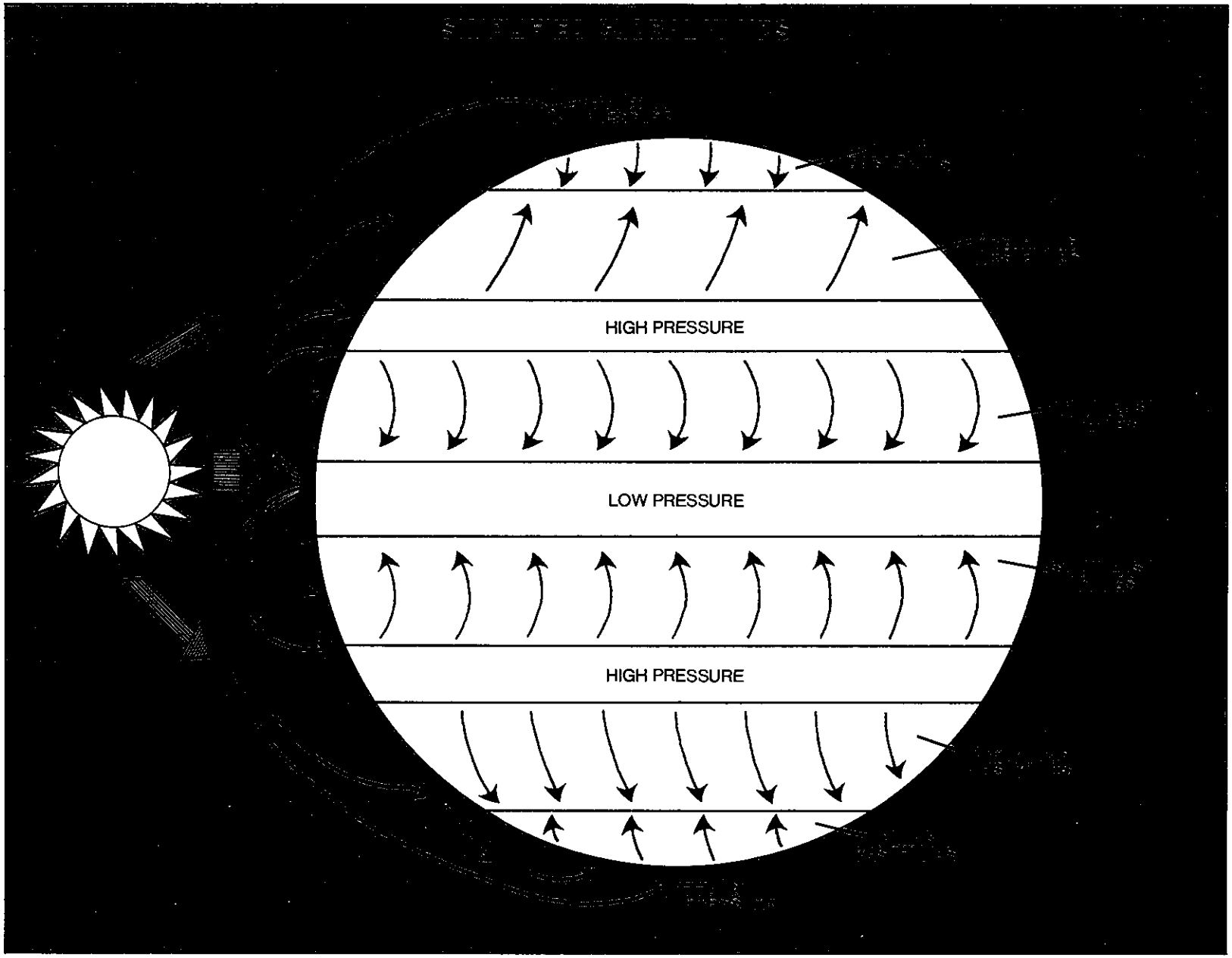
The global circulation (worldwide wind pattern) of wind flow is arranged in three broad belts in each hemisphere: 1) the *North and South Easterly Trades* are found in the Tropics and Subtropics, 2) the *prevailing westerlies* are found in the middle latitudes, and 3) the *Polar Easterlies* are found in the higher Arctic and Antartic latitudes.

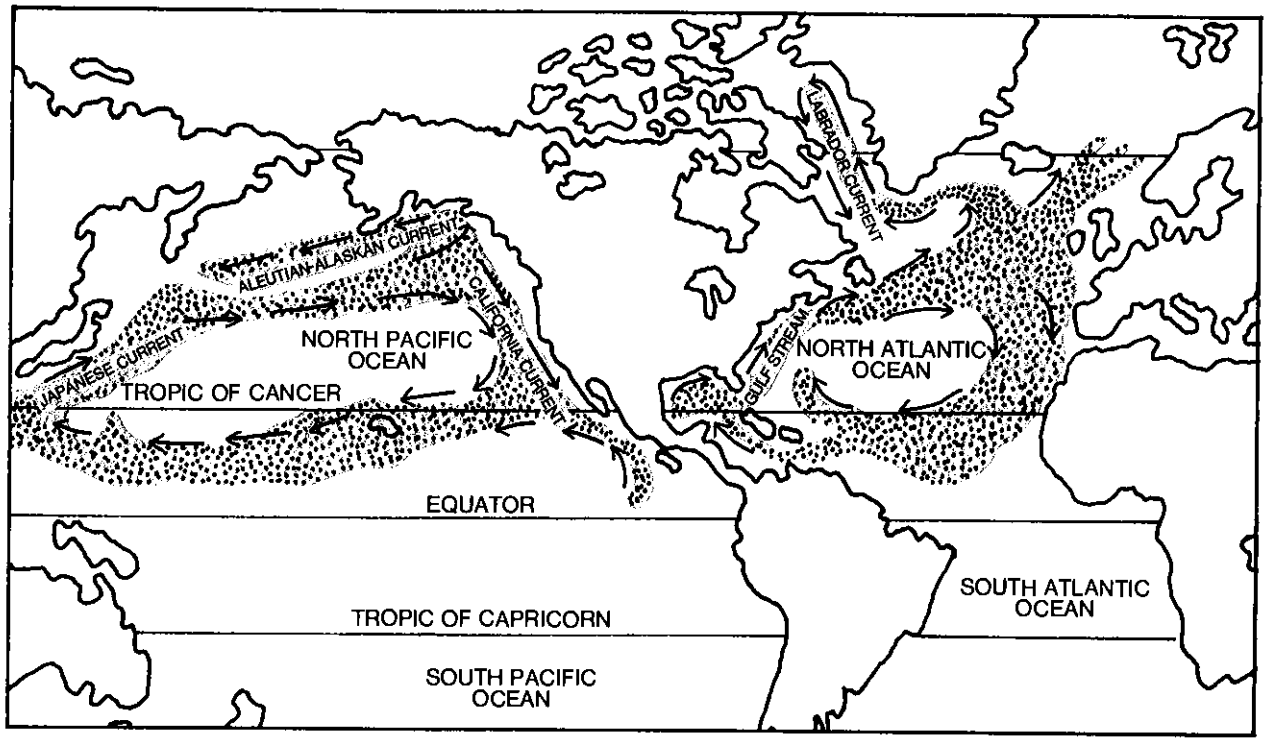
The rotation of the earth about its axis causes the wind to be deflected to the right in the Northern Hemisphere and to the left in the Southern Hemisphere. (For example: In the Northern Hemisphere a wind blowing from the south is deflected toward the east and a wind from the north is turned toward the west). This deflection is called the *Coriolis Effect.*

High pressure areas are the results of the formation of large air masses called Anticyclones which rotate clockwise in the Northern hemisphere. Between these high pressure areas are cyclones (low pressure areas) which rotate counterclockwise in the Northern hemisphere. Highs are usually homogeneous with temperature and humidity being much the same throughout the air mass. Cold dry highs move into the United States from Canada traveling generally southeastward.

Fronts are usually formed along the line between two different air masses. Because of temperature and humidity differences, the air masses do not mix readily usually causing active or turbulent weather.

Ocean Currents: Coastal areas are influenced by well-established ocean currents. The best known perhaps, is the Gulf Stream, which carries warm waters through the Florida Straits, northward along the Atlantic Coast to Cape Hatteras, and then northeast and eastward to the British Isles. Without the Gulf Stream, Great Britain, Ireland and Western Europe would have a much colder climate—more typical of their latitude. Winds blowing across this warm current, bring heat and moisture to the land, both on the south coast of the United States and in western Europe. The climate of the northeastern coast of the United States is affected by another current, the cold Labrador current which flows southward as far as Virginia.


HIGH PRESSURE
LOW PRESSURE
HIGH PRESSURE



SOME MAJOR OCEAN CURRENTS AFFECTING COASTAL WEATHER OF THE U.S.

The Japanese current is the major factor affecting the weather of the West Coast of the United States. After passing the shores of Japan, it branches to become the Aleutian — Alaskan Current bringing a milder climate to coastal portions of Alaska and Canada, and the California current which brings cooler waters to California and the lower west coast of the United States.

Elevation (Altitude): Air-temperature in the atmosphere decreases with increasing elevation. The usual rate of decrease, called *Lapse Rate,* is 3.3°F per 1000 ft.

Mountain Barriers: Mountain ranges not only have colder temperatures at higher elevations but also act as barriers to physically block the flow of prevailing winds and storm movement. Also, as the air flows upward on the slope of a mountain barrier the air mass lifts much of the moisture and releases it on the windward side of the range as it cools. On the leeward side of the range arid and semi-arid conditions usually exist.

Florida: Florida, surrounded by water on all sides but the north, does not have as great an annual range in temperature as many inland states do. It does retain a high humidity year-round. Southern Florida has a sub-tropical climate and winter polar fronts seldom penetrate to the Miami area. However, freezing temperatures and frost which accompany cold air masses reach northern and central Florida nearly every year. In other seasons, the weather of the entire state is more or less similar — warm and humid. The central region exhibits a high frequency of thunderstorms in the summer (among the highest in the nation). Also, Florida has been swept by hurricanes more often than any other state. Some of these originate in the tropical Atlantic, others in the Gulf of Mexico, the maximum hours of sunshine (solar radiation) received yearly.

TO THE TEACHER

There are many exercises which can be done to develop a knowledge of meteorology. In fact, the study of weather should be treated as a complete course which it is in several schools. But the fact that weather needs to be considered when doing field exercises, or when studying the marine environment is why it is mentioned here.

When observing the changes in ecological systems, collecting water quality data, or studying any marine organism, the weather conditions at the time of study must be taken into consideration. Students should keep record of meteorological conditions and discuss them in the analysis of their other observations. All that is intended here, is that students learn that there is a very definite relation and close interaction between the water (including all that lives within) and the atmosphere above it.

NATIONAL WEATHER SERVICE

The Surface Weather Map — Its Purpose and Use

Nearly all weather maps read by the National Weather Service are prepared at the National Meteorological Center in the Washington D.C. area. From this location at least 100 weather charts, serving a multitude of purposes, are distributed daily to 350 weather stations in the United States.

At the National Weather Service Office station weather charts are received on a facsimile machine. This piece of electronic equipment greatly helps your local National Weather Service personnel know what is going on weather-wise and why it is happening. The chart information comes to

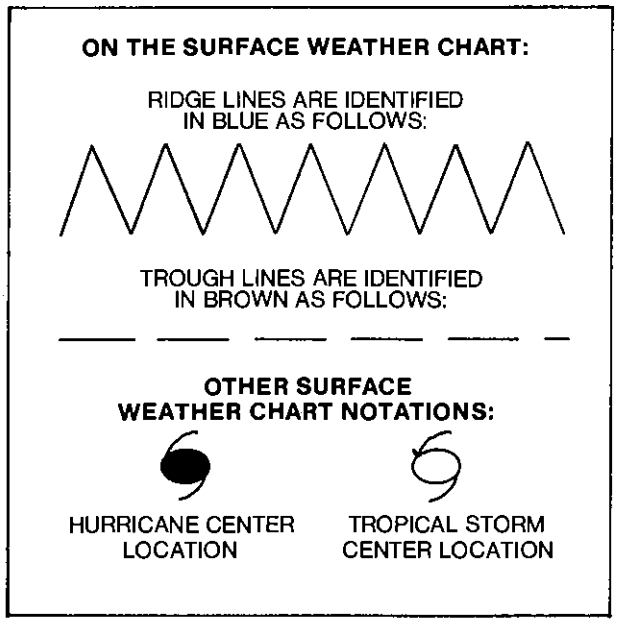
us through the use of a map transmitting unit in the Washington D.C. area which photoelectrically scans the original data. These electrical impulses are then picked up in a news station on a special telephone line which is connected to the facsimile machine receiver. In short, the receiver burns the transmitted information onto premoistened paper, one line at a time. It takes about 20 minutes to obtain a copy of the latest surface weather map, which is the largest chart we receive.

Surface weather maps are prepared every 3 hours from simultaneous weather observations taken at about 500 locations in North America. These observations are entered on a base map at the National Meterological Center and analyzed by a meteorologist for atmospheric pressure and fronts. The resulting surface analysis chart, as it is also referred to, gives the user a quick picture of the weather across the nation for a specific time. The National Weather Service recently developed the Automation of Field Operations and Services (AFOS) program. With this new system, scheduled for completion in 1980, the Weather Service will accumulate, process, translate and disseminate weather data faster. The system incorporates advanced computer technology and CRT (cathode ray tube) screen display to eliminate the printing of surface maps on paper. The meteorologist will be able to make faster, more accurate forecasts.

The time used on all weather charts is based on a 24 hour clock and is given in Greenwich Mean Time (Z); i.e. the time in Greenwich, England, which is on the zero degree meridian. Please note the time entry in the lower left hand corner of the chart. To convert this time to Eastern Standard Time, subtract 5 hours. If we are on Eastern Daylight Time, subtract 4 hours.

One of the most obvious features of the surface analysis chart is its multitude of numbered lines. These are called isobars and connect all points having the same atmospheric pressure. The metric system of millibar units is used to label all isobars, with the lines drawn at a 4 millibar interval.

Example: Isobar Label
96 = 996 millibars
00 = 1000 millibars
04 = 1004 millibars



Note . . . 1000 millibars is equivalent to 29.53 inches of mercury.

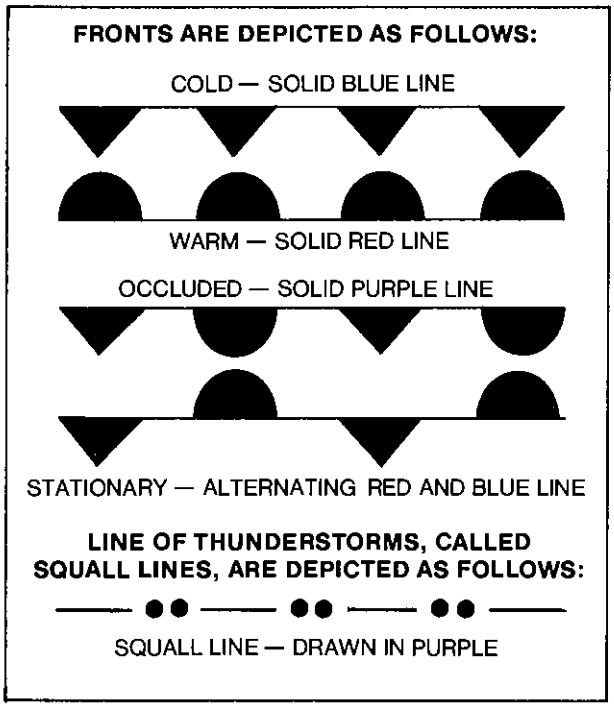
The closer two or more isobars are next to each other, the steeper the pressure the gradient, and the stronger the wind will be blowing.

The highest or lowest pressure with these centers is indicated by the underline numbers near the H or L.

Example: 99 L = Lowest pressure of 999 millibars
H 42 = Highest pressure of 1042 millibars

Isobars on a surface weather chart can be compared with height contour lines on a topographic chart. The highs being like mountain peaks or ridges and the lows being like sink holes or valleys.

Fronts are very important features of the surface analysis and their location is identified on the chart by the meteorologist. Across a front the atmospheric conditions change drastically; i.e. temperature, moisture, wind direction and speed, pressure change, precipitation and cloudiness. Generally, fronts emanate out of low pressure centers and continue across the weather chart in pressure troughs (valley area).



Individual plots of the weather, from which the chart is analyzed, is also available for the user to see what is occurring at a specific location.

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CHARTS AND NAVIGATION

INTRODUCTION

The word navigate is taken from Latin "navigere" meaning *navis* (ship) *agere* (move or to direct). Marine navigation may be defined as "process of directing the movement of a craft from one point to another on or in a body of water." According to our best estimates man has been navigating the ocean for at least 6000 years — some say perhaps 8000 years or more. Most impressive were the Polynesians, who traversed thousands of miles of open ocean in the Pacific using charts of reeds as early at 1500 B.C.

Among the most important instruments developed to aid navigation of the sea are the compass, chronometer and the sextant. Date and origin of the magnetic compass are unknown. It is one of the oldest intruments. A gyroscope tends to retain a given direction and in this century we devised the gyrocompass that is independent of magnetic fluctuations. The chronometer, an extremely accurate clock is used to determine longitude. Remember, if the earth is viewed from the pole, it rotates through 15° each hour ($24 \times 15° = 360°$). Then each 15° of east or west travel on earth represents one hour's difference. (Refer to Time Zones such as Eastern, Central and Pacific). The sextant was invented about the 17th or 18th century. This instrument enables a mariner to determine location at sea with excellent accuracy. The electronic age has added radar, loran and sounding equipment (sonar).

A *map* is a representation of the earth's surface on a plane surface — a *chart* is a map used primarily for navigation. A nautical chart intended primarily for marine navigation shows land outline, prominent land features (natural and man-made), depth of water, channels, navigation markers, a compass rose and other aids to navigation.

To represent or project the surface of the earth (a spheroid), on a flat surface will cause some distortion. All maps have some distortion, therefore there are different projections for different purposes.

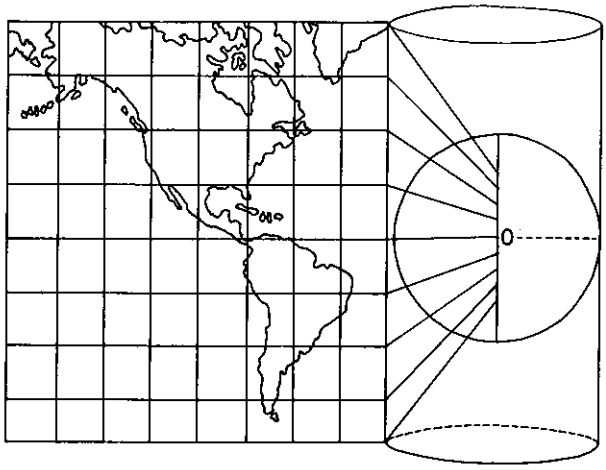
Desirable properties of a chart are to represent:

1) the true shape of physical features
2) the correct angular relationship
3) area in correct relative proportions
4) constant scale values for measuring distances

No one projection has all of these properties, therefore every chart has some distortion. Charts of small areas of earth, such as the map of a harbor, will have practically no distortion and can be used without corrections.

Perhaps the best known projection used is the *Mercator,* named for Geradus Mercator, a Flemish geographer, who designed and published it in 1569. There are any number of other projections including conic, azimuthal, polar and gnomonic to name a few. Of these, gnomonic is believed to be the oldest — thought to have been developed by the Greek, Thales, about 600 B.C.

MERCATOR CONFORMAL PROJECTION



Today, chart making is generally categorized among the sciences. It is called *cartography.* Several federal agencies are involved in conducting surveys, preparing data, drafting and printing charts. Substantial cartographic research is underway at most of these agencies which include: N.O.S. (National Oceanic Survey), USGS (US Geological Survey), DAM (Defense Mapping Agency), FAA (Federal Aviation Agency), NASA (National Aeronautical and Space Administration), Lake Survey, Mississippi River Commission, and U.S. Army Corp of Engineers. Each contributes to the chart making process in its own special areas.

TO THE TEACHER

There are standard symbols used on all nautical charts. Copies of Chart No. 1, Nautical Chart Symbols and Abbreviations. USA can be obtained from the U.S. Coast and Geodetic Survey, Department of Commerce, Washington, D.C., or from the local chart dealer or book store. Also obtain some charts for student use. Not all charts have to be of your area, but try to get one or more of the area you may visit on a field trip. Intracoastal Waterway charts are excellent for these exercises. In Exercise I, students will become familiar with chart symbols. In Exercise II, Plotting a Position, the following activities are recommended: 1. Students can practice with a hand-bearing compass by sighting objects in the classroom or the schoolyard. Have students navigate from object to object returning to the point of origin. 2. On a chart,

select objects and various bearings and have students practice "walking" parallel lines from the compass rose to the objects. (Any lines marked on the chart should be light and in pencil). 3. Have students find a position on a chart in the classroom by giving them three bearings, e.g. marker #4 bearing 40°, radio tower WFOS bearing 50°, and city pier bearing 90°. 4. Plan a sampling field trip in which students plot their location even if they are not on board a boat. For Exercise III, give students a series of fictitous courses, speeds and times. Also introduce a fictitous current or wind and ask students to determine their estimated position.

TO THE STUDENT

Exercise I — CHART SYMBOLS

MATERIALS

Chart No. 1
Nautical chart of your local or neary area

Using the chart, locate the following objects or conditions on your chart.

1. The compass rose
2. A can buoy
3. A nun buoy
4. A green daymark
5. A red daymark
6. A lighted buoy (what color?)
7. A fixed bridge (vertical clear?)
8. A draw bridge (specify type)
9. A wreck
10. A radio tower
11. A landmark (what?)
12. a) accurate position
    b) approximate position
13. A storage tank
14. A dredged area (depth and date)
15. A spoil area (two types of bottom)

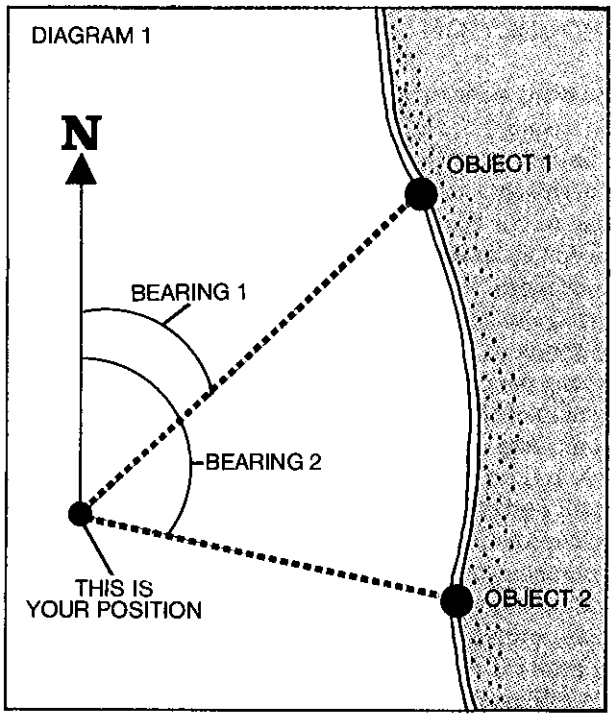
Exercise II — PLOTTING A POSITION

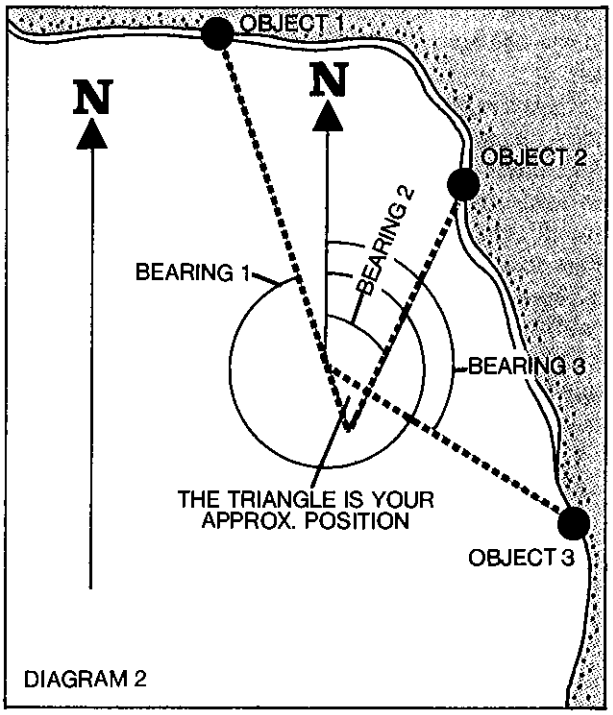
MATERIALS

Chart of the area
Pencil
Plotting device (parallel ruler or drawing triangles)
Hand-bearing compass.

When sampling data or collecting is done, it is necessary to record the location where the data was collected. At times this is quite simple, such as when you are at the end of a certain street or dock or the middle of the Main St. bridge. Suppose you are sampling from a boat in a river or bay, with a hand-bearing compass and chart of the area. You can "fix" your position, chart your course, and identify the sampling sites just as accurately as before.

At the site, consult the chart for two or three permanent objects that are both within sight and marked on the chart. Using the hand-bearing compass, sight on one of these objects and record its bearing from you. Locate that bearing on the compass rose and "walk" a set of parallels until the line goes through the object sighted. Mark this line lightly on the chart. Repeat these steps for the remaining objects. These lines will cross at a point or make a small triangle. Your location is where the lines cross.





By using three bearings, error is reduced and the fix is the center of the triangle formed.

Exercise III —DEAD RECKONING

There may be conditions such as night or fog, where one will not be able to see many objects to establish a fix. If you must move or cannot hold your position under these circumstances you must rely on dead reckoning. Dead reckoning is the process of determining your position by using only your last known position and the course and speed of the boat.

If these conditions occur, first note your last recorded position and calculate your distance traveled. For example, your last known position was sea buoy 18. Your boat traveled at a speed of 10 knots for 30 min. on a heading (compass course) of 135° (one knot is one nautical mile per hour). Next calculate your distance traveled in nautical miles by multiplying the boats' speed in knots times the hours traveled. Mark this distance on the line. This is your Dead Reckoning Position. Anytime dead reckoning is used be alert for any objects that will confirm a position or location (sound, light, breakers, smells etc.).

What effect would winds and currents have on your position?

QUESTIONS FOR CONSIDERATION

1. Why are three bearings more desirable than two for locating a station?
2. Why are symbols used on chart? Why not write everything out?
3. Why must dead reckoning be used?
4. Why is it important to find a position accurately?
5. Where did the term "knots" come from?

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BATHYMETRIC ANALYSIS

INTRODUCTION

On nautical charts, water depths are an aid to navigation. Reefs, submerged pilings and other obstructions are also noted.

Suppose you wanted to chart a body of water and include depths and obstructions on your chart. Where would you begin? First you would need a map showing the outline of the body of water. Then you would need to go out on the water and measure the depth at various locations, being sure to record your exact location on the water. Now, by recording these depths at the corresponding location on your map, you will have constructed your chart. This type of chart is called a bathymetric chart, a chart of depths.

Black Box - A Model Exercise of Bottom Profiling
A Simulated Preparation of a Bathymetric Chart

TO THE TEACHER

The Black Box is prepared in advance. The box is constructed from one half sheet of ¾ inch exterior plywood, and sixteen feet of 1 by 12 boards*. This material will make a box that measures 4 feet by 4 feet by 1 foot. Fill the bottom of the box with bricks, broken block, wood scraps and other objects of various size and shape to form an uneven profile. Then fill the rest of the box with clean builder's sand.

Students probe the depths with a stiff wire, measuring the depth of the water (sand) by noting the length of the wire that penetrated the sand. The student then records the data on a chart and graphs a profile of the unseen floor.

This exercise simulates the techniques used by oceanographers. It will give the student an idea of the difficulty of mapping the unseen ocean bottom accurately.

Assign a pair of students to track their "ship" across the ocean (box). The profile developed by each team will reveal the topography under the track but is only part of the information needed for a map of the whole bottom. When all teams have completed an ocean track the topography of the ocean bottom can be shown.

**One 4' x 4' sheet of ¼" pegboard. Optional pegboard as a top cover provides a grid system. The holes can be identified as latitude and longitude or some other numbering or lettering system.*

TO THE STUDENT

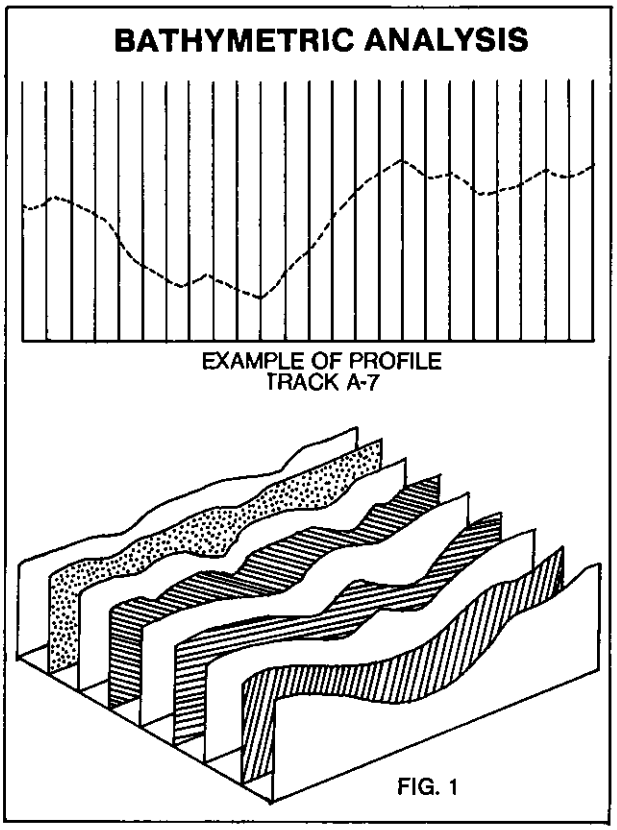
Often one looks at a bathymetric chart and accepts it as being 100% accurate, not realizing the work involved and how much is really unknown. Bathymetric charts are the result of many soundings and are as accurate as the data collected permits. The more soundings actually taken, the more accurate the chart. Mariners, ecologists, marine researcher, submariners and many others are vitally interested in the topography of the oceans for ship safety and navigation. To provide an impression of the oceanographer's work "making a bathymetric chart", you will probe and chart the profile of a track across the sea (box).

PURPOSE

To demonstrate the technique in surveying the ocean to prepare the bathymetric chart.

MATERIALS

Box 4' x 4' x 1' filled with sand and rock
Stiff wire — suggest clothes hanger or brass welding rod
Metric rule
Line to mark track
Paper to record results and make chart



PROCEDURE

1. Designate teams (2 students each)
2. Assign teams and determine an actual track
3. At specified intervals along the track insert the wire probe vertically to measure depth of sand to rock
4. Record position and depth
5. Make a scale drawing of the profile

After all teams have produced their profiles, develop a compoite bathymetric chart as accurately as possible in class as an activity. Further activity: To visually illustrate the topography, make profiles out of construction paper. See figure.

QUESTIONS FOR CONSIDERATION

1. How does the size of the interval between stations affect accuracy?

2. How can inconsistent features, i.e. isolated peak, one great depth, be explained? How could one check these inconsistent features for accuracy?

3. Does one of a few tracks really show the general topography?

4. How important is acurate navigation in sailing the seas? How can bathymetric charts be used for nagivation?

5. Are accurate charts important in the deep ocean areas? Explain your answer.

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CORROSION AND GALVANIC ACTION

INTRODUCTION

Even the most casual observer can identify corrosion on metal items seen everyday in the city or country. In the auto junk yard, in port and harbor areas, in the farmyard and in structural building activities of cities; in short, wherever steel or iron is in use, one can see evidence of the oxidization process underway. The oxide of iron is commonly known as *rust.*

Corrosion is considered to consist of the slow chemical and electro-chemical reactions between metal and its environment. Essentially all reactions of metal in the atmosphere (oxygen & water vapor) or in electrical conducting liquids (electrolites, which are aqueous solutions of salts, acids or bases) are electrochemical in nature.

Different metals are affected to different degrees and corrosive attack takes many different forms. Attack may be made by 1) general *tarnishing or rusting* with occasional perforations. 2) localized *pitting* attack 3) *crevice* attack, and 4) *intergranular* attack. Two special types of intergranular attack are *grooving* and *layer* corrosion.

Corrosion may proceed at a rapid or slow rate. The rate is controlled by the metal undergoing attack and by the environment, the concentration of the reactants and the temperature. Environmental conditions greatly influence the nature and extent of damage to metals. The form of attack could be of a general nature covering the whole surface or it might be very localized and specific in nature. The many variables and factors involved in the corrosive environment need to be analyzed and understood as fully as possible. For example, metals immersed in sea water may suffer increased damage by the attachment of certain marine organisms or, at shore locations wave action, strong currents and tidal action (which alternately covers and exposes materials) are factors which promote the rate of deterioration.

Galvanic corrosion is a special type of metallic deterioration that occurs when two dissimilar metals are immersed in an electrolyte. It is of interest to the mariner and marine engineers because ships, pilings and offshore platforms often utilize different metals underwater. Sea water is a good electrolyte, allowing a current to flow between the two metals. The result is that the more active metal (according to the galvanic series tables) at the anode is corroded away, and the less active metal at the cathode is unaffected (protected) (see fig. 1).

TO THE TEACHER

Corrosion, particularly corrosion of iron and steel is a phenomenon that most students become acquainted with early in their childhood. An opportunity exists in this series of exercises to establish a basic foundation of knowledge on how corrosion takes place. Perhaps it would be worth while to reiterate the chemical principle: matter can neither be created or destroyed. It changes form through chemical activity.

Research is continually underway in industry and government agencies to investigate the corrosion process, and how to reduce or prevent it.

Be sure to differentiate between corrosion and erosion. The first is an electrochemical process which changes the metallic element to a compound (oxide, chloride, sulfides . . .) the other is a change in physical characteristics. On the other hand, erosion is a mechanical process by which metal is removed by abrasion. In addition, the physical and chemical characteristics of the metal are unchanged even though the shape of the object is altered.

Have students bring in various corroded metals. Students may be able to identify *pitting, stress corrosion, crevice corrosion, layer corrosion* and *intergranular corrosion.*

TO THE STUDENT:

Each day we see corrosion of metal surfaces around us. Probably the most common is rust (iron oxide). It is frequently observed on a car, used tin can, on a wire fence or maybe a nail. The green "stain" one observes on a copper sheeted church dome, or copper rain downspout is corrosion, as is the whitish substance on aluminum screens. All corrosion is a natural process which is at work to return the metal to its original natural state as an ore. Man's changing of the ore to a metal is thus only temporary.

There is a hydrological cycle which regenerates water for various uses and an ecological cycle which reduces dead organisms to nutrients to provide continuous supply of food for plants and animals; the process of corrosion is, in many respects, similar. Could you prepare a simplified diagram of the refinement/corrosion process of a metal? Is it a cycle? What name might it be given?

EXERCISE I

PURPOSE

To examine the corrosive activity of metals in sea water.

MATERIALS

Strips of copper (Cu)
Zinc (Zn), Iron (Fe), Lead (Pb), Platinum (Pt) and calcium (Ca) turnings tied to a string.
Covered glass containers
Sea water
Metric balance

PROCEDURE

1. Cut the metals into nearly equal sized strips. Weigh each strip and record.
2. Suspend the strips or turnings in the containers so that they will be half-way submerged in the sea water. Label.
3. Cover the container to retard evaporation.
4. Examine each container hourly the first day and daily thereafter for two weeks. Record the results.

OBSERVATIONS AND CALCULATIONS

1. Take the mass of each metallic object at the end of the two weeks after cleaning the attached corrosion from the strip (omit the calcium turnings).
2. Divide the mass of the original strip of metal into the mass of the remaining strip after the experiment. Multiply by 100. This is the percent of the metal remaining.
3. Arrange the percentages from step 2 in descending order.
4. If the sea water changed color, note as observed.

QUESTIONS

1. Which metal would you use to make underwater fittings?
2. Did the metals corrode equally underwater?
3. Was the metal affected at or above the water line? If so describe.

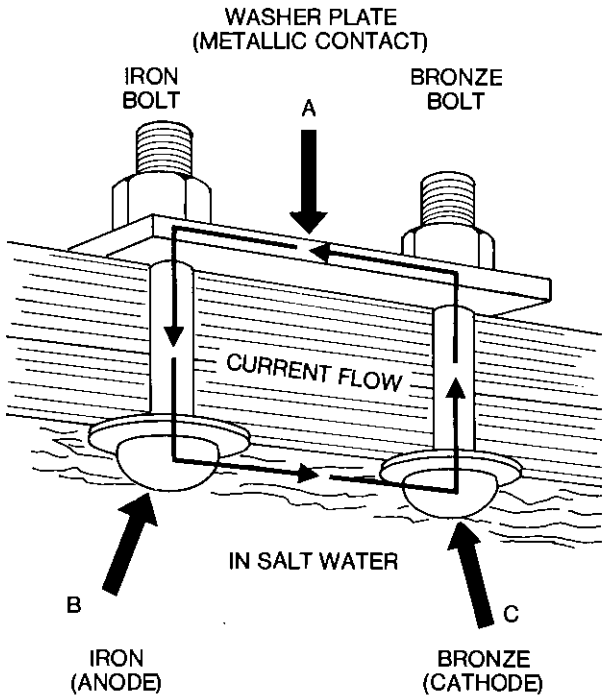
SUGGESTED FOLLOW UP EXERCISE

Upon completion of Exercise I, immerse the metals totally, then suspend over the water so that all the metal is exposed to the air.

Immerse again each day for a week and record your observations.

QUESTIONS FOR CONSIDERATION

1. Did the corrosion process speed up or slow down with exposure to the air?
2. What part does the salt play in this process?



SIMPLE
CORRECTIVE MEASURES

A—Break connection.
(by using separate washers)

B—Add sacrificial protectors to anode.

C—Replace either bolt with one made of metal that is closer on the galvanic scale to the metal of the other bolt.

GALVANIC SERIES

least noble—anodic
(corroded end)

MAGNESIUM
ZINC
ALUMINUM
CADMIUM
STEEL OR IRON
CAST IRON
STAINLESS STEEL
(active)
LEAD
TIN
MANGANESE
BRONZE
BRASSES
COPPER
(COPPER PAINT)
SILICON BRONZE
(EVERDUR)
MONEL
NICKEL
(passive)
SILVER
GRAPHITE
GOLD
MERCURY
(MERCURY PAINT)

most noble—cathodic
(protected end)

EXERCISE II

PURPOSE

To examine the corrosive nature of various solutions (electrolytes).

MATERIALS

Copper strips of known mass
Sea water
*HCL solution
*HNO_3 solution
*H_2S solution
*H_2SO_4
*NH_4OH
Closed containers
Balance
*Suggestion—buy these premixed in solution or have it done in the chemistry dept.

PROCEDURE

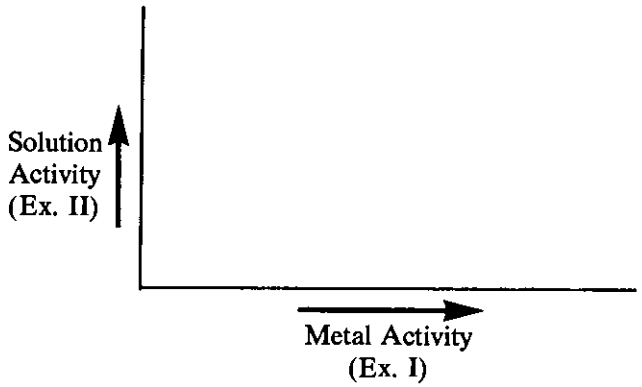
1. Measure each solution into a separate container. Mark the liquid level.
2. Suspend the copper strips in each solution (half in, half out). Loosely cover.
3. Observe bubbles and color change. Continue observations hourly for the first day and daily thereafter for two weeks.
4. Remove the strips. Rinse to remove corrosion and reweigh.

OBSERVATIONS AND CALCULATIONS

1. Measure the mass of each strip two weeks after removing corrosion.
2. Divide the mass of the original strip of metal into the remaining mass after the experiment. Multiply by 100. This is the percent of metal remaining.
3. Arrange the percentages from step 2 in descending order. What does this tell you about the rate of corrosiveness of the liquids.

QUESTIONS FOR CONSIDERATION

1. Which liquids changed colors? Why?
2. Compare the two exercises. Prepare any x-y chart as follows:



Consider the upper right portion of the graph where the most active metal/solution cross. What does this tell you about the combination?

ALTERNATE EXERCISES

1. Coat one half of each strip with various coatings (chromeplate, paint, vaseline or nail polish) and repeat the experiments. How do the results change?
2. Try an experiment on the order of #2 but instead of a single copper strip, use a pair of strips clipped together at the top. Try Copper/Zinc, Iron/Zinc, Aluminum/Iron for comparison. The difference in results is caused by galvanic action (see fig. 1).
3. Can galvanic action or electric currents be utilized to build structures under sea water by using the electric current to precipitate the sea minerals?

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THE USE OF UNDERWATER PHOTOGRAPHY IN A MARINE SCIENCE CLASS

Marine science teachers are finding it increasingly difficult to provide field experience in the natural environment. Spreading urban development is displacing our natural ecosystems and increasing the distance to suitable field sites.

All too many of our students have little experience with or knowledge of their own local natural environment. Even students who have lived in the area all of their lives are not aware of their natural surroundings.

If the teacher cannot get the students into the field, photography offers an effective tool to bring the environment to the student. Commercial audio-visual products have been used for years, but they often fall short because of their failure to relate to the local area. 35mm slide programs, produced by the teacher, can be very effective teaching aids, providing students with an overview of the natural ecosystems making up the local area, as well as providing a visual survey of the flora and fauna inhabitating those areas.

TO THE TEACHER

Experience has shown some added benefits that occur from teacher-made slide programs.

1. Taking the pictures improves the teacher's power of observation and naturally lends itself to a better understanding of the ecosystem or subjects being photographed.
2. Slides taken and used by the teacher even if of lesser quality than professional slides, will help establish respect for the teacher's personal effort to improve the class. The students generally respond to this kind of effort on the part of their teacher.
3. Students will relate much better to slides of local flora and fauna than those from areas remote to them. Native scenes will also reinforce their own personal observations.
4. Slide programs enliven a classroom routine, providing more variety of classroom techniques.
5. Plant and animal classifications can be surveyed, using local examples in their natural setting, rather than using killed and preserved specimens. Often our collecting just adds to the stresses on diminishing local populations.
6. Test questions using the slide visuals can be very effective. Identification quizzes can be given using slides.
7. Macro-photography isn't difficult, and can show details that the students might otherwise miss.
8. Often students interested in photography will participate in making slides and programs and from this participation will learn some marine science in spite of themselves.
9. A few scenic programs put to music by slide-tape synchronization adds another dimension to the teacher's classroom techniques.

EQUIPMENT SELECTION

In photography, you can spend a great deal of money especially in acquiring underwater equipment. Unfortunately, most schools and teachers have limited funds, so we will limit this discussion to practical, relatively inexpensive basic equipment needs. Since we are dealing with subjects related to marine biology, we will assume the need is for underwater photographic equipment.

A good quality 35mm single lens reflex camera or the Nikonos 35mm amphibious viewfinder camera has proven to be an excellent starting point. It's very doubtful that you will get satisfactory results with the smaller pocket cameras. If you or your school can acquire both of the above mentioned cameras, you will have the basic tools to start producing useful slide programs. If you have your choice of only one of the two, some difficult choices must be made.

Most of today's moderately priced SLR Cameras take excellent quality slides. Compared to the price of accessories for the amphibious camera, the SLR can be a versatile tool. With a normal 50mm lens and the addition of a few inexpensive extension tubes for macro-photography, you can produce a variety of marine-oriented programs. But obviously, unless you house the camera in a water-tight case, you are limited to the surface. Here again we are faced with some difficult choices. Some of the choices are expensive. If you are able to acquire one of the Nikonos model amphibious cameras, you will have an extremely versatile compact camera underwater and an optically excellent 35mm lens camera on the surface.

Underwater equipment manufacturers have produced a large variety of lenses ranging from dome port wide angles to telephoto to macro and close-up lenses. There are also several strobe and flash units, filters, extension cubes with framers, optically corrected viewfinders, convenient attachment handles and brackets, thumb-controlled shutter levers, and light meters.

If you own or have one of the popular brand SLR cameras, chances are someone makes a production model housing for it. Most of the modern housings are reliable and well designed.

If you have the proper tools and the necessary skills, you can make your own housing, but even the materials and specialized parts aren't cheap. Mort Loggweilers book, *How to Build Your Own Underwater Camera Housing* is available from Hydrotech Co., Box 1444, Long Beach, Calif. 90814.

The July, 1979 issue of Skin Diver Magazine contains a report on the testing of a flexible housing called EWA Marine. The report was favorable if the photographer limits

himself to shallow water because only available light can be used. The price presently ranges from $49.00 to $85.00, depending on the model camera being used.

When you become proficient with either type of camera, with available light in shallow water, you may want to acquire an artificial light source, such as an electronic strobe or an underwater flash attachment. Artificial light enables you to capture the beautiful colors found in deeper water that would otherwise be filtered out by the column of water above.

Once again, you are faced with some difficult choices concerning expense vs. convenience and quality. The less expensive underwater flash attachment offers an excellent light source, but the cost per picture is high, due to the expense of flash bulbs. The electronic strobe requires a higher initial financial investment, but a lower overall cost over a few years of use. Rechargeable strobes are available on models using replaceable batteries.

Another option is housing a surface strobe. Some manufacturers make housings for some models of surface SLR strobes. If you should already have one of these models, it will reduce your initial costs.

Whatever your choice, the purchase of an artificial light source can wait until you become proficient in available light photography. By then, you can better determine your future needs for artificial light.

FILM

All currently available films are excellent. However, for a place to start we would recommend a film with an ASA 64 as a general purpose film. Some of the newer films with ASA's of 200 or 400 have been producing excellent results for low-light conditions.

FILTERS

To compensate for the loss of red light rays and to overcome the "blue haze" in deep blue or green waters, utilize the CC30R filter to improve and enhance natural colors. In yellowish-tinted water you might try a CCM filter (Magenta) to improve the color.

While it is beyond the scope of this article to delve into the techniques and the more technical aspects of underwater photography, Jim and Cathy Church have a monthly series in *Skin Diver Magazine* that is an excellent resource for the underwater photographer. Joe Strykowski's book *Divers and Cameras* is a comprehensive text on the subject.

SOME CARE AND MAINTENANCE TIPS WORTH REPEATING

Saltwater and heat are enemies of camera equipment and film. It would be wise to use something like a styrofoam ice chest to store film and loaded camera while exposed to heat and bright sunlight.

After every use, your underwater equipment should be washed in mild, soapy fresh water, then well rinsed. After the equipment is dry, it should be disassembled and carefully inspected for sand, salt particles and corrosion. External metal parts can be given a light coat of silicon to protect their finish. All o-rings should be inspected and greased, then carefully stored for their next use.

SLIDE ORGANIZATION

As soon as you start producing slides for use in your classroom you would be wise to develop your own system for classification, labeling and cataloging. Start with your very first slide and continue with each thereafter. If you delay, you run the risk of falling hopelessly behind in this time-consuming task. It is well worth the time to develop a key-code to use for each slide. Indicate what it is, where it belongs, and how it can be used.

Example: a picture of a branch of staghorn coral belonging to the Phylum Cnidaria. Your code for this phylum is the code number 7. So you place a 7 on the slide frame. You could, if you wished, place a capital A after the 7 to indicate it belongs to the Class Anthozoa. An M after the A indicates it is from the order of stony corals called Madreporaria. You could then place another number to indicate in what order that slide was exposed and chosen for your program. Now the code reads 7AM18, if it was the 18th slide you chose for classroom use.

A master list of your slides becomes more and more valuable as your collection grows. As your slides begin to pile up, it is important that you have an aide in helping you choose your slide subject, locate them, and set up your program.

A light board of some sort is necessary to help you set up your programs and edit your slides. Light boards are inexpensive to buy or can easily be made.

Inexpensive plastic boxes can be used to store slides not in trays, or to store duplicate slides set aside for use in other programs.

It may seem unnecessary, but you would be wise to prepare a check list to avoid forgetting anything on your next filming trip. How often trips are ruined because something vital is left behind. Before you depart, check off each item as it is placed in your boat or car. Believe it or not, the item most often left behind is film. Film and batteries are often stored in the home refrigerator and too many times are still there while the photographer is sitting dejectedly on the boat.

Since slides are vulnerable to damage and loss it is recommended that duplicates of the slides normally used be made. This way the originals will be safe and in original condition.

One scheme you might get some ideas from, is as follows:

SLIDE SUBJECT	CODE	DATE	LOCATION TAKEN	EXPOSURE INFO	STORAGE LOCATION	IDENTIFICATION
STAGHORN CORAL ACROPORA CERVECORNIS	7AM18	7/13/79	FRENCH REEF	F-8a+60	BOX 7	CORAL REEFS PHYLUM CNIDARIA

DETERMINING THE AGE OF TELEOST FISH BY COUNTING SCALE RINGS

INTRODUCTION

In future years it may become very important to know the age composition of each type of fish. Through knowledge gained about the amount of growth attained each year and the life span of fishes, man can harvest the sea crop with greater efficiency.

Length-frequency distribution is the oldest age determination method in use. This involves large numbers of specimens and is based on the theory that all fish of one size are approximately the same age. This method has built-in disadvantages in that: (a) it does not separate older age groups, which tend to remain approximately the same size; (b) fish of the same size have a tendency to school together; (c) variable environmental conditions may allow one age group to grow at a faster rate or a reduced rate each year; (d) hatching does not alway occur at the same time of the year; (e) it requires a large number of fish in a random sample.

The most positive and effective method of determining the age of fish is through the marking, release, and recovery of fish of a known age. The major disadvantages of this method are (a) cost of tagging; (b) time imvolvement; (c) the small number of specimens recovered; and (d) damage from handling.

A generally accepted method of determining the age of fish is to count the annual layers deposited in the hard parts of a fish. The best hard structure to count is the scale, while the ear stone (otolith) and spine are secondary because of location in the specimen. This method is dependent upon several items: (a) accuracy upon the individual's interpretive ability, and (b) upon the distinctiveness of the annual layers. These layers in the hard parts of fishes are produced by metabolic processes and occur in a similar manner to the growth rings produced by trees each year.

TO THE TEACHER

In selecting specimens for use in this exercise, only fully-formed scales should be selected. It must also be realized that scale markings of fish in the tropical zone are sometimes imperfect because of the lack of a true winter season to slow the growth process. Sometimes a false annulus is produced by females just prior to spawning as reabsorption occurs.

It may be to your advantage to select scales from large specimens for easier student observation — Snook, Bluefish, Jewfish, Sea Trout, Tarpon, etc. Taxidermists may be a good source of scales for study.

Scales can be stored for use by placing them in an envelope or pressed between pieces of paper. Mucus on the scale will hold the paper together. Species, place, weight, length, sex, collector, time and method of capture should be recorded with the scales.

The scales can be placed between two layers of thin, clear plastic and mounted in a 35mm slide blank and projected upon a screen to produce the necessary magnification. This preparation lends itself to easy storing and cataloging.

Other methods of examination would include the overhead projector, opaque projector, and a light box.

Some fish (catfish and eel) do not possess scales or the markings on the scales are not clear enough for interpretation. In these cases, the otolith is used. The otolith is a calcareous structure formed in the inner ear of the fish. Some thin otoliths can be read directly, others must be sectioned. In fact, some researchers also recommend grinding and polishing before interpretation.

TO THE STUDENT

In attempting to determine the age of fishes by studying the structure of the scale, there are several definitions which must be given before starting:

Annulus —The annual mark or zone found on the scales, vertebrae, otoliths or other hard portion of a fish, which is formed once a year. (Figure 2).

Ctenoid scale — The scale of a bony fish that possess small sharp spines (ctenii). (Figure 1)

Cycloid scale — The scale of a bony fish without spines or ctenii. (Figure 1)

Ganoid scale — Thick plate-like scales. The annual rings are not well defined.

Focus — The small clear area near the center of a scale that represents the original scale of the young fish. (Figures 1 and 2)

The definitions given for cycloid and ctenoid are not fixed. There are many variations or degrees of spination or stenii and the position of focus varies with each species of fish.

The annulus is recognized in one of the following ways: (a) "crossing over" where with the onset of fall or winter several circuli ridges flare outward and end on the side of the scale rather than circle the focus; (b) "discontinuous circuli" — the individual circuli do not grow together in a complete line because the scale stops growing; and (c) extreme crowding of the circuli which occurs just prior to a resumption of growth. (See Figure 2)

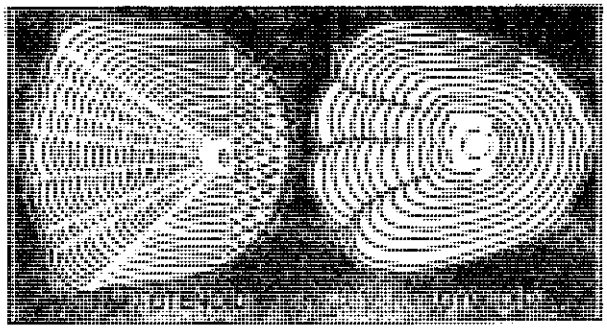

FIG. 1

PURPOSE

To determine the age of a bony fish by examination of its scales.

MATERIALS

Dissecting microscope or hand magnifier
Slides
Fish of several sizes or scales provided by the instructor
Metric rule
Projector
Light box

PROCEDURE

1. Remove a scale from each specimen to determine whether it is cycloid, ctenoid or ganoid.
2. If the scale is ctenoid, take scales for further examination from the region of the pectoral fin; cycloid scales should be removed from an area between the dorsal fin and the lateral line. (Rounsefell and Everhart, 1953)
3. Remove 3 scales from the indicated area from each specimen.
4. Mount the scales between two glass slides for observation under the microscope.
5. Record your observations.

ORGANIZED DATA RECORDING

1. Prepare a line drawing illustrating the general features of the scale: focus, annuli, circuli, ctenii (if present).
2. Using your metric ruler, determine the distance between the annuli on the scale (repeat for each scale of each specimen).
3. Each scale should be read twice, at different times, in order to arrive at an accurate interpretation of structure.
4. Counting one year of growth for each annulus, determine the age of each specimen.

QUESTIONS FOR CONSIDERATION

1. How can you account for the varying distances between each annulus on the scales?
2. How much difference occurs in the distance between annuli on scales from the same specimen? Why does this occur, if present?
3. How is age determination of fishes useful in fisheries biology?

LIMITATIONS AND SOURCES OF ERROR

1. Incorrect reading of scales.
2. Use of imperfect scales or scales that have been regenerated.
3. Some fishes show no definite annuli.
4. Errors in age determination increase with the age of the fish.
5. Errors made in determining the location of the first annulus.

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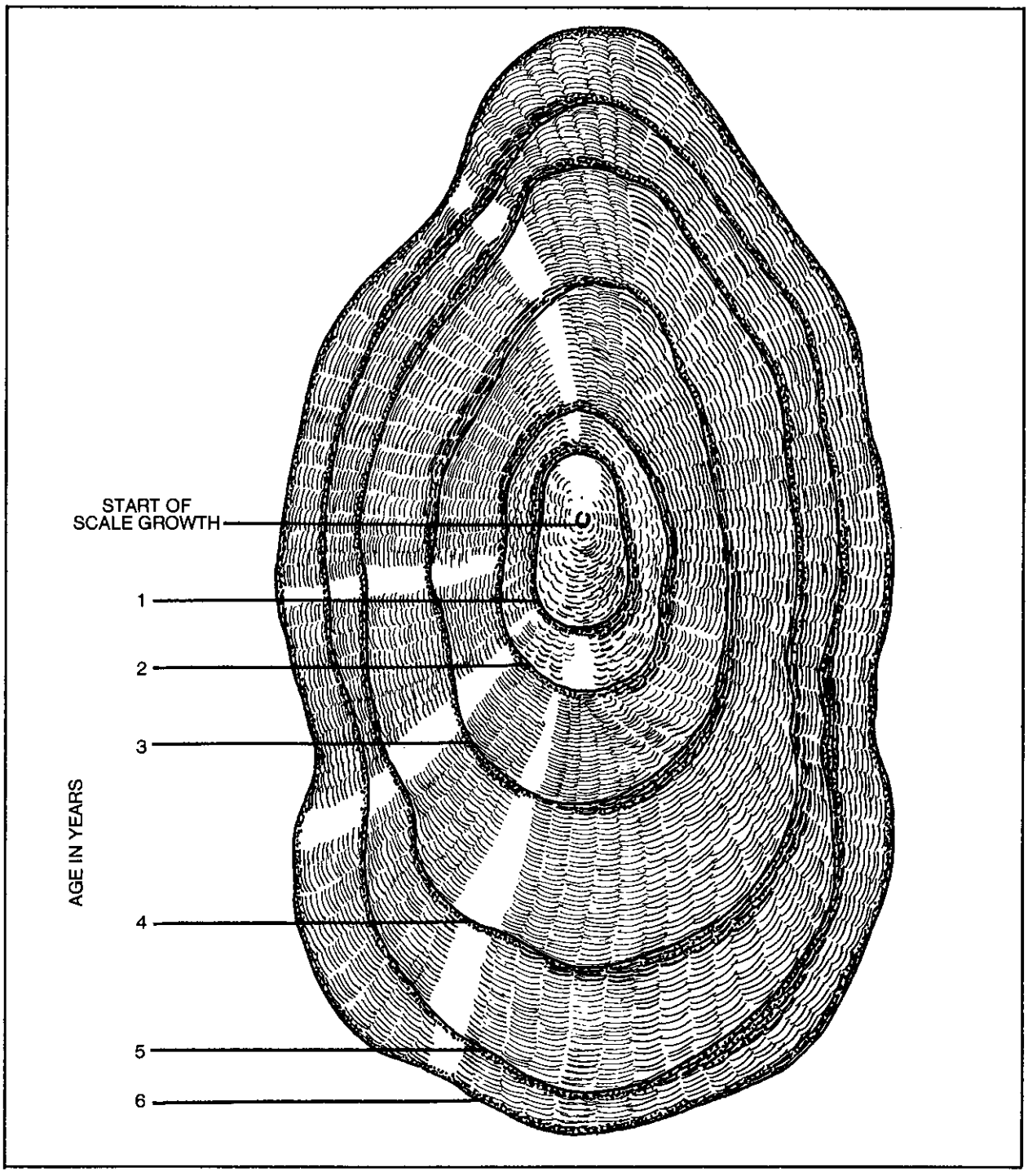


FIG. 2 Typical cycloid scale enlarged from a 6 year old haddock. The more closely spaced rings (circuli) form darker year marks (annuli). The total annuli count determines the age of the fish. (U.S. Bureau of Commercial Fisheries.)

FIELD TRIPS

- The School Time Field Trip
- Planning
- After School or Weekend Trip
- General Suggestions for Teachers
- Safety Parameters for Field Trips
- Suggested Ecology Field Trip Letters

"Science" might best be considered and defined as the "act of discovering and reading the natural world." True, conceptual schemes (theories) are developed and many facts of quantifiable data are gathered and considered very important in relating to each concept. Yet if "science" is to be considered a verb, it requires mental and physical action. The best approach to science, then, is to *do* science. Science activities in the field lend themselves well to developing personal values, more so than in a typical conventional classroom situation.

When formulating goals for marine science courses, include with conceptual schemes and process themes, personal values to be developed. The best means of carrying out your value goals are through the following: (1) laboratory group experiments, (2) colloquiums following lab activities, (3) slide-tapes, (4) projects and assignments; and (5) field trips. In marine sciences important values should include a respect for the living organisms of the sea and a commitment to maintain a healthy habitat for them.

How do field trips differ from controlled laboratory experiences?

(1) Serendipity method of inquiry (with prepared mind) vs. orderly arrangement of process tools. Both are valid "methods" in obtaining scientific information and the field trip lends itself more to the serendipity method.

(2) Field trips enable students to see that real situations are results of several variables and that closed-systems really exist only in theoretical physics. Controlled experiments carried out in the extreme could yield an over-simplistic view of the natural world.

Marine biology is still mostly a field-oriented subject, at least in the minds of most students, and much of its intrinsic appeal lies in field work. Yet you, the teacher of marine biology, may have some serious doubts about your personal ability to run a good, safe, constructive learning experience in the field. Worse yet, you may not be permitted to take students into the field on school time.

Take heart! You are not alone and field experiences are not only possible under adverse circumstances, but are also usually positive happenings for all concerned.

In order to collect in the marine environment, one must obtain a permit issued by the Florida Marine Patrol. This permit enables the science instructor to collect marine organisms (except endangered species) provided the collecting is not done within an aquatic perserve, a Florida State Park or any of the federal refuges or sanctuaries. In most instances, the permit is restricted to certain counties. Be certain that you contact the local office of the Marine Patrol, an agency of the Florida Department of Natural Resources, before going on a beach, marsh or coastal field trip.

THE SCHOOL-TIME FIELD TRIP

Once you have decided a field trip is a necessary part of your instruction, or a positive addition to a unit, several advance preparations need to be made.

PLANNING

ONE MONTH AHEAD

Make a first scouting trip to the site.
Discuss the trip with your principal.
Start "talking it up" in the class.
Start collecting needed equipment.

TWO WEEKS AHEAD

Permission slips go out to students.
Transportation arrangements roughly finalized.
Start setting up student teams.

ONE WEEK AHEAD

Revisit the site (to be sure it hasn't become a building site!).

Collect permission slips from students.

Start of list of students who will go.

Check with principal to be sure nothing has been left undone (last minute surprises generally have a negative effect).

Hand out to the students checklists, behavior rules and other necessary paperwork. If you want students to use these at a later date either make enough to distribute again or collect and redistribute at appropriate time.

THE DAY BEFORE

Organize equipment and students for transport.

Double check transportation and other vital items. (Substitute, permission slips, etc.)

Finalize list of students for tomorrow's roll call.

THE BIG DAY

Turn in list of students on trip, keep a copy for yourself.

Count heads before leaving school and before leaving field site.

Be mentally ready for unexpected happenings or changes, including a usually sleeping student becoming the best investigator and an "A" student being totally confused.

Obviously we haven't covered all eventualities with this quick survey, and more thoughts on useful details will be found at the end of this section.

THE AFTER-SCHOOL OR WEEKEND TRIP

Legal or other considerations may make a school-time trip impossible. In this case, a voluntary trip for interested students (and their families) may be the answer, generally conducted on a weekend. It may be possible also to operate trips under the auspices of an organization, such as Explorer Scouts, Junior Audubon, YMCA, or YWCA or a local church in order to obtain insurance protection for teacher and participants. In any case, the rough schedule outline previously described will help in setting up the trip. The basic differences between this volunteer "after-hours" trip and a school-sponsored one include:

- not all students will be able to attend;
- transportation is usually family car rather than school bus;
- the trip emphasis is more enrichment than rigorously academic;
- insurance coverage requires a different approach; look into "camper" insurance;
- people other than your students will also attend.

This type of trip does have several advantages, including the ability to do an overnight trip and a much relaxed, "fun" atmosphere.

GENERAL SUGGESTIONS FOR TEACHERS

- Adhere strictly to local regulation, both pertaining to students and to the site.
- Do you have permission to enter the property?
- Do you have your collecting permit?
- Have appropriate agencies been notified?
- In general, make it a policy that students may *NOT* drive, even if they are over 18; this avoids many problems.
- Lecturing in the field is very difficult; therefore, individual student worksheets are preferable.
- Do you have a "rain plan"?
- Safety is your responsibility. Be dogmatic about adherence to safety rules.
- A camera and tape recorder provide excellent post-trip material. Keep the microphone very close to the person speaking. A cardboard funnel around the mike will focus sounds, including voices, birds, the slurping of feet.
- Use the metric system. "What's the temperature of the water in degrees Celsius?"
- Scavenger hunts, show and tell, find the elusive organism, are all useful organizing helps.
- Bring along appropriate field guides, let students identify organisms.
- Remember to remain ecologically sane: don't allow overcollecting, return organisms alive to their home and replace overturned rocks.
- Use other adults as teacher aides by giving them written assignments; some as "border guards," some as student team assistants, etc. Try not to let "turned on" adults monopolize your time.
- Ask other adults who went on the trip to comment constructively. Often a very supportive letter results, which helps future trips.
- Prepare students ahead of time with slides, maps, written materials which describe and define the site, its ecology, history, problems.
- Student teams of two or three work well. If there are more than three to a team then one or more student will "slack off."

SAFETY PARAMETERS FOR FIELD TRIPS

Your responsibility in the field is all-encompassing. Insurance coverage should be settled before you plan a trip. Beyond that your main concern is to warn students (in writing) ahead of time of potential hazards, to set down (in writing) rules of behavior, and then to insist on absolute adherence to your rules.

1. Procure insurance coverage through your school or sponsoring organization.
2. Require one adult for every 10 students.
3. Allow only responsible adults who are not students to drive. Your best bets are other school personnel or parents; be especially wary of friends of students or brothers and sisters of students. A chartered bus, school or otherwise, is the safest and preferred means of transportation.
4. Provide a written set of safety rules and regulations to each participant and adult. For out-of-school-time trips, require students to return to you a copy of rules and regulations signed by a parent before the trip.
5. Allow *NO* infractions; before the trip, plan a method for dealing with students who do not adhere to safety rules.
6. Go over your safety precautions with your superior or with another "field-wise" teacher to catch any omissions.
7. Marine trips generally require a few additional precautions, such as:
   a) no one will be allowed to wade without wearing shoes that tie (sticky mud!).
   b) to avoid stingrays teach students to slide their feet along the bottom without lifting them.
   c) specify the depth to which you will allow wading; generally knee-keep is more than sufficient.
   d) no swimming unless a card-carrying lifeguard is available, and on duty for every ten swimmers.
8. A first aid kit is mandatory. Buses are required to carry them. The kit is useless unless someone it trained to use it — preferably you. Include some meat tenderizer or papaya juice for treatment of protein based "stings" from marine organisms, e.g. *Physalia* (Portuguese Man-of-War), Jellyfish and Sting Rays.
9. Where is the nearest hospital? How would you get there from the site?

SUGGESTED ECOLOGY FIELD TRIP LETTERS

The following are two examples of letters to the parents concerning field trips. Each school system may have an already established policy so it may be best to check that first.

Example I

Dear Parent,

As you probably know, your son or daughter is enrolled in the course entitled "ECOLOGY-12." The true laboratory for such a course is the out-of-doors. Therefore, several field trips are planned for the year. There will be an average of one to three local field trips per week and one "non-local" field trip per six week period. Often we will need to call on a student to assist with transportation since buses are needed to take other students home. If for personal reasons, health reasons, or economic reasons a student wishes to miss a field trip, he may do so. The local field trips may last until 4:30. Any student is free to attend all field trips, however, none are compulsory.

As for conduct on the field trips, students are to conduct themselves as gentlemen/ladies. They are to remember that they represent members of the student body. Field trips are usually fun, BUT are intended primarily as learning experiences. For longer trips, food/lunches will be the responsibility of the student. Gas costs and any admission costs will be shared by the entire class (those attending).

If your son or daughter receives permission from you to attend a field trip, I will try my best to make it a meaningful educational experience. Please note, however, that full responsibility for learning rests on the student (and the parent). Learning is minimal if discipline requires excessive time and attention. Problem situations (individuals) will not be allowed on future field trips and firm action may be taken to insure welfare of the students.

Yours truly,

.....

I have read the above letter and agree to allow my son/daughter _____ to attend the field trips under the conditions stated. I relieve the _____ School of all responsibility.

Date _____ Signature _____

Example II

GENERAL INFORMATION FOR YOUR TRIP
TO
THE ENVIRONMENTAL STUDIES CENTER

1. ALL STUDENTS MUST WEAR SHOES AT ALL TIMES, PREFERABLY TENNIS SHOES. THONGS ARE NOT ALLOWED FOR ANY TRIPS.

2. WEAR SUITABLE CLOTHES. IF YOUR GROUP IS GOING SEINING OR IN THE WATER, YOU SHOULD ALSO BRING A CHANGE OF DRY CLOTHES AND A TOWEL, (NO BATHING SUITS), AND NO SWIMMING!!

3. ALL SCHOOL RULES APPLY THE ENTIRE TIME YOU ARE AT THE CENTER OR ON THE BOAT. THIS INCLUDES NO SMOKING.

4. BRING YOUR OWN LUNCH AND SOFT DRINK. THERE ARE NO LUNCHES OR DRINKS AVAILABLE AT THE CENTER OR ON THE BOAT. YOU MAY WANT TO HAVE SOMEONE IN YOUR CLASS BRING A SMALL COOLER WITH ICE FOR YOUR SOFT DRINKS.

5. LEAVE RADIOS AND TAPE PLAYERS AT HOME.

6. FOR STUDENTS GOING OUT ON THE BOAT:

   YOU WILL HAVE TO WEAR A LIFE VEST THE ENTIRE TIME YOU ARE ON THE BOAT. THERE ARE NO EXCEPTIONS.

   IF THE WEATHER IS COOL IT WILL BE MUCH COOLER IN THE WIND ON THE BOAT. BRING A JACKET AND WEAR LONG PANTS OVER YOUR SHORTS.

7. IF WE HAVE TO CANCEL YOUR TRIP BECAUSE OF BAD WEATHER WE WILL RE-SCHEDULE YOU AT THE EARLIEST POSSIBLE DATE.

Martin County Schools'
Environmental Studies Center
2900 NE Indian River Drive
Jensen Beach, FL 33457

MARINE SCIENCE CENTERS AND SOURCES

- National Organizations and Agencies
- Other Sources of Marine Science Information
- Directories
- Research Organizations and Agencies in Florida
- Sea Grant/Marine Advisory Program
- Environmental Education Centers in Florida

National Organizations and Agencies

American Fisheries Society, 5410 Grosvenor Lane, Bethesda, Maryland 20014, 301/897-8616.

Bureau of Outdoor Recreation, U.S. Department of the Interior, Division of Information, Washington, D.C. 20240, 202/343-5726.

Federal Water Pollution Administration, U.S. Department of the Interior, Washington, D.C. 20240, 202/337-2500.

Fish and Wildfish Service, U.S. Department of the Interior, 18th & C St., N.W., Washington, D.C. 20230, 202/343-5634.

National Marine Education Association, Curriculum Committee & Project Coast, 310 Willard Hall, University of Delaware, Newark, Delaware 19711, 302/738-2333.

National Marine Fisheries, U.S. Department of Commerce, Washington, D.C. 20235, 202/634-7281.

National Oceanic and Atmospheric Administration, (NOAA), U.S. Department of Commerce, National Ocean Survey, Rockville, Maryland 20852, 301/436-6980 or 443-8186.

National Oceanic and Atmospheric Administration, (NOAA), U.S. Department of Commerce, Office of Coastal Zone Management, 3300 Whitehaven Street, N.W., Washington, D.C. 20235, 202/655-4000.

National Wildlife Federation, 1412 16th Street, N.W., Washington, D.C. 20036, 202/797-6800.

National Youth Fishing Program, AFTMA Center, 2625 Clearbrook Drive, Arlington Heights, Illinois 60005, 312/364-4667.

Superintendent of Documents, U.S. Government Printing Office, Wasington, D.C. 20400, 202/783-3238.

U.S. Coast Guard, U.S. Department of Defense, Washington, D.C. 20373, 202/426-2158.

U.S. Naval Oceanographic Office, U.S. Department of Defense, Washington, D.C. 20373, 202/545-6700.

National Climatic Center, Federal Bldr., Asheville, N.C. 28800.

National Oceanographic Data Center, Information branch, Page Bldg., Washington, D.C. 20235.

National Hurricane Research Lab., Univ. of Miami, 1365 Memorial Drive, P.O. Box 8265, Coral Gables, Florida 33124.

Joint Tsunami Research Effort, U. of Hawaii, 2525 Correa Rd., Honolulu, HI 96822.

SpaceCraft Oceanography Group, NOAA/NESS, Suitland, MD 20023.

Atlantic Estuarine Fisheries Center, Pivers Is., P.O. Box 570, Beaufort, N.C. 28516.

Gulf Coast Fisheries Center, Bldg. 302, Ft. Crockett, Galveston, TX 77550.

Atlantic Marine Center, 439 W. York St., Norfolk, VA 32508.

National Weather Service, Silver Spring, MD 20910.

National Hurricane Center, P.O. Box 8286, Coral Gables, FL 33124.

U.S. Army Corps. of Engineers, Jacksonville District, P.O. Box 4970, Jacksonville, FL 32201.

U.S. Army Corps. of Engineers, Mobile Dictrict, P.O. Box 2288, Mobile, AL 36628.

Coastal Engineering Research Center, Kingman Bldg., Ft. Belvoir, VA 22060.

Defense Mapping Agency, Hydrographic Center, Washington, D.C. 20373.

Environmental Protection Agency, Public Affairs Office, Washington, D.C. 20460, *Region IV* — 1421 Peachtree St., N.E. Atlanta, GA 30309.

NASA, Washington, D.C., National Space Data Center, Goddard S.F.C. Greenbelt, M.D. 20770.

Dept. Transportation, Washington, D.C., *U.S. Coast Guard Oceanographic* Unit Bldg., 159-E, Navy Yard Annex, Washington, D.C. 20390.

Coastal Plains Center for Marine Development Services, 1518 Harbour Drive, Wilmington, N.C. 28401.

Gulf States Marine Fisheries Commission, 531 St. Louis St., New Orleans, LA 70130.

Marine Mammal Comm./Council on Environmental Quality, 712 Jackson Place N.W., Washington, D.C. 20006.

Other Sources of Marine Science Information

Lamont-Doherty Geological Observatory, Columbia Univ., Torrey Cliff, Palisades, N.Y. 10964.

Scripps Institution of Oceanography, La Jolle, CA 92037.

Marine Biological Lab., 1056 MBL St., Woods Hole, MA 02543.

Woods Hole Oceanographic Institution, Woods Hole, MA 02543.

Wrightsville Bio-Medical Lab, 7205 Wrightsville, Ave., Wilmington, N.C. 28401.

Bernice P. Bishop Museum, Pacific, Science Infor. Center, P.O. Box 6037, Honolulu, HI 96818.

Marine Technology Society, Suite 412, 1730 M St., N.W., Washington, D.C. 20036.

Smithsonian Science Infor. Exchange, Rm. 300, 1730 M St., N.W., Washington, D.C. 20036.

Smithsonian Oceanographic Sorting Center, (Biological Specimens) Bldg. 150, Navy Yard Annex, Washington, D.C. 20390.

Oceanographer of Navy, Information Division, 200 Stovall St., Alexandria, VA 22332.

Under-Sea Medical Society, 9650 Rockville Pike, Bethesda, MD 20014.

Directories

Directory of: Development in The Marine Environment of the Coastal Plains Region, issued annually by: Coastal Plains Center for Marine Development Service, Wilmington, N.C. 28401.

Marine Science Newsletters—1973, An annotated Bibliography, NOAA Tech. Memo Eds Nodc-3, Issued by National Oceanographic Data Center, Washington, D.C. 20235.

Directory of Newsletters, Sea Grant Marine Advisory Service, Issued by Sea Grant Office/NOAA, Washington, D.C. 20235.

Undersea Technology Handbook Directory, Issued by: Compass Pub. Inc., Arlington, VA.

University Curricula in the Marine Sciences and Related Fields, Issued biannually by Sea Grant Office/NOAA, Washington, D.C. 20235.

A Directory of Florida's Environmental Education Centers, Issued by: Office of Environmental Education, W.V. Knott Bldg., Tallahassee, FL 32301.

Directory of Marine Education and Research Facilities in Florida, Florida Sea Grant University of Florida, Gainesville, FL 32611.

Research Organizations and Agencies in Florida

The Universities that do research in the marine field are:

Florida State University
University of South Florida
University of West Florida
Florida Atlantic University
Florida International Union
University of Florida
University of North Florida
University of Miami

U.S. Geological Survey, Water Resources Division, 325 John Knox Rd., Suite F-240, Tallahassee, FL 32303, 904/386-1118.

Naval Coastal Systems Center, Panama City, FL 32401, 904/234-4011.

National Marine Fisheries Service, SEFC, Panama City Laboratory, 3500 Delwood Beach Rd., Panama City, FL 32407, 904/234-6541.

National Marine Fisheries Service — Southeast Region, 9450 Koger Blvd., St. Petersburg, FL 33702, 813/893-3141 or 826-3141.

National Oceanic and Atmospheric Administration, National Marine Fisheries Service, Southeast Fisheries Center, 75 Virginia Beach Drive, Miami, FL 33149, 305/361-5761.

NOAA Atlantic Oceanographic and Meteorological Laboratories, 15 Rickenbacker Causeway, Miami, FL 33129, 305/361-3361 ext. 350/353.

U.S. EPA Environmental Research Laboratory, Gulf Breeze, FL 32561, 904/932-5311.

U.S. Coast Guard, 7th Coast Guard District, 51 S.W. First Ave., Miami, FL 33130, 305/350-5641.

United States Department of Agriculture Soil Conservation Service, P.O. Box 1208, Gainesville, FL 32602, 904/377-8732.

Florida Department of Natural Resources Marine Research Laboratory, 100 Eighth Avenue S.E., St. Petersburg, FL 33701, 813/896-8626.

West Florida Arthropod Research Laboratory, P.O. Box 2326, Panama City, FL 32401, 904/785-6159 or 769-0201.

Aqualife Research Corporation, P.O. Box 3414, Marathon Shores, FL 33052, 305/289-1550.

Battelle Columbus Laboratories, Florida Marine Research Facility, Sailfish Drive, Ponce Inlet, Daytona Beach, FL 32019, 904/761-3072 or 767-3330.

Columbia Research Corporation, Gulf Coast Division, Coastal Resources Analysis Branch, P.O. Box 9453, Panama City, FL 32407, 904/234-8817.

General Oceanics, Inc., 5535 N.W. 7th Ave., Miami, FL 33127, 305/754-6658

Gulf Specimen Co., Inc., P.O. Box 237, Panacea, FL 32346, 904/984-5297.

Gulfarium, Highway 98 East, Fort Walton Beach, FL 32548, 904/244-5169 or 242-8378.

Harbor Branch Foundation, Inc., RR 1, Box 196, Fort Pierce, FL 33450, 305/465-2400

Institute for Delphinid Research, P.O. Box Dolphin, Marathon Shores, Florida Keys 33052, 305/289-1121.

Marineland, Inc., Rt. 1, Box 122, St. Augustine, FL 32084, 904/471-1111.

Mote Marine Laboratory, Inc., 1600 City Island Park, Sarasota, FL 33577, 813/388-4444.

Ocean Farming Systems, Inc., P.O. Box 164, Tavernier, FL 33070, 305/852-3624/9284.

Ocean World, Incorporated, 17th St. Causeway, Fort Lauderdale, FL 33316, 305/525-6612.

Potomac Research, Incorporated, 1607 Lisenby Avenue, Panama City, FL 32405, 904/769-3352.

Tropical Bioindustries, 900 Southwest 87 Court, Room 104, Miami, FL 33176, 305/279-7026.

Sea World of Florida, 7007 Sea World Drive, Orlando, Florida 32809, 305/351-3600.

Wometco Miami Seaquarium, 400 Richenbacker Causeway, Miami, FL 33149, 305/361-5705.

Associated Marine Institute, Inc., 1311 N. Westshore Blvd., Ste. 202, Tampa, FL 33607, 813/879-7137.

Dade Marine Institute, 400 A-Rickenbacker Causeway, Miami, FL 33149, Miami Seaquarium, 361-7934.

Florida Keys Marine Institute, Young Adult Conservation Corps, P.O. Box 1116, Key West, FL 33040, 305/294-5119.

Florida Marine Science Education Association, President-elect: Marjorie R. Gordon, 6301 Summit Blvd., West Palm Beach, FL 33406, 305/686-6600.

Florida Ocean Sciences Institute, Inc., 3563 N.W. 8th Ave., Pompano Beach, FL 33064, 305/942-1120.

Florida Oceanographic Society, 1212 Riverside Drive, Stuart, FL 33494, 305/287-1950.

The International Oceanographic Foundation, 3979 Rickenbacker Causeway, Virginia Key, Miami, FL 33149, 305/361-5786.

Jacksonville Marine Institute, 1825 E. 21st St., Jacksonville, FL 32206, 353-7555 or 353-7556.

Marine Science Education Center, 1347 Palmer St., Mayport, FL 32233, 904/246-2733 or 246-1521.

Maritime Agricultural Training Experience, P.O. Drawer 1430, Key West, FL 33040, 305/294-4773.

Museum of Science, Inc., 3280 S. Miami Ave., Miami, FL 33129, 305/854-4242.

Panama City Marine Institute, 222 E. Beach Drive, Panama City, FL 32401, 904/763-0748.

Tampa Marine Institute, 1310 Shoreline Drive, Tampa, FL 33605, 813/248-5091.

Florida Marine Life Association, 156 Dove Ave., Tavernira, FL 33070, 305/852-5459.

Florida Shore & Beach Preservation Association, Inc., 325 John Knox Rd., F-214, Tallahasse, FL 32303, 904/386-1983.

Florida Waterways Association, Inc., P.O. Box 1766, Palatka, FL 32077, 904/328-5869.

Gulf of Mexico Fishery Management Council, 5401 W. Kennedy Blvd., No. 881, Tampa, FL 33607, 813/228-2815.

Jacksonville Port Authority, P.O. Box 3005, Jacksonville, FL 32206, 904/633-5247.

The Marine Council of Greater Miami, 615 S.W. 2nd Ave., Miami, FL 33130, 305/856-0206.

Tampa Port Authority, P.O. Box 2192, Tampa, FL 33601, 813/248-1924.

Environmental Science and Engineering, Inc., 14220 Newberry Real, P.O. Box 13454, Gainesville, FL 32604, 904/372-3318.

Witney Marine Research Laboratory, University of Florida, Route 1, Box 121, St. Augustine, FL 32804, 904/829-5607.

Coastal Data & Engineering, Inc., 2038 N.E. Ridge Ave., Jensen Beach, FL 33457, 305/334-2722.

Sea Grant/Marine Advisory Program

The Marine Advisory Program is a part of the state's Sea Grant program which provides information and help in the various fields of marine related topics throughout the state. The Advisory Agents may cover a particular region of the state and can be a valuable source of information to local teachers.

In Florida there are at present 7 Marine Advisory Agents. For information about the Florida Marine Advisory Program, write or call:

Dr. Marion Clarke, Coordinator
Marine Advisory Program
120 Newins — Ziegler Hall
University of Florida
Gainesville, FL 32611
904/392-1837

Environmental Education Centers in Florida

Listed here are some Environmental Education Centers in Florida which have activities in marine science.

Marine Sciences Under Sail
Post Office Box 3994
Hollywood, Florida 33023
305/983-7015

Marine Science Center
P.O. Box 1258
Crystal River, FL 32629
904/795-4393

Marine Science Education Center
1347 Palmer Street
Mayport, Florida 32233
904/246-2733

Dade County Environmental Education Center
150 West McIntrye Street
Key Biscayne, Florida 33149
305/350-3506

Interpretive Section, Dade County and Recreation Dept.
50 Southwest 32nd Road
Miami, Florida 33129
305/854-3530

Hillsborough Community College's Environmental Studies Center at Cockroach Bay
Post Office Box 22127
Tampa, Florida 33622
813/879-7222

Hobe Sound Nature Center
U.S. Fish and Wildlife Service
Hobe Sound, Florida 33455
305/546-6141

Florida Audubon House
Post Office Drawer 7
Maitland, Florida 32751
305/647-2615

Pine Jog Environmental Sciences Center
6301 Summit Boulevard
West Palm Beach, Florida 33406
305/395-5100 or 686-6600

Nature's Classroom
Route 1, Box 396
Thonotosassa, Florida 33592
813/986-1867

Jacksonville Children's Museum, Inc.
1025 Gulf Life Drive
Jacksonville, Florida 32207
904/396-7061

S. Bryan Jennings Environmental Center
9719 West Beaver
Jacksonville, Florida 32220
904/781-1434

Environmental Studies Center
207 East Main Street
Pensacola, Florida 32501
904/438-1140

Martin County School's Environmental Studies Center
2900 N.E. Indian River Drive
Jensen Beach, Florida 33457
305/334-1262

Lee County Environmental Education
Center/Nature Center
2055 Central Ave.
Fort Myers, Florida 33901
813/334-1983

Newfound Harbor Marine Institute
Route 1, Box 170
Big Pine Key, Florida 33043
305/872-2331

Energy Management Center
Post Office Box 190
Port Richey, Florida 33568
813/848-4870 or 848-4881

For further information write to:
Office of Environmental Education
Department of Education
W.V. Knott Build
Tallahassee, Florida 32301

TIPS FOR TEACHERS

- Bio Art
- Bio-Mounting Fish and Other Marine Species
- Fresh Water at the Seashore
- How to Make an Undersea Thermometer
- Aerial Photos For Free
- Data File System
- Quick and Easy Method for Brine Shrimp
- Invertebrate Aquarium
- Collecting
- Binder/Perry Sampler

BIO ART

If students leave your classroom at the end of the year and never again become involved with the environment, they may forget most of what they have learned. A teacher must strive not to teach marine science as a segregated subject but as a subject that can be intergrated with many aspects of the student's life. Not every student will be science oriented when leaving school. Thus, try tying together marine science and art experiences.

Two individuals are missing something exciting:

(1) The pure marine scientist who fails to stop and look at the beauty of this environment.

(2) The artist that sees the inherent beauty in the marine environment and uses it in artistic expressions but fails to understand any of the science of the area nor even know the name of the "friends" that he paints, sculptures or photographs.

Try this bio-art project. Have a *Bio-art Fair Day,* requiring the student to submit an art project dealing with the sea along with a research report concerning the organisms or physical parameters involved. Give the student at least two months advance notice. Let them know that the project will count a great amount towards their grade and failing to do the project might lead to a failing grade. Let them know that the project will be graded as to the time and concern obviously put into it not on the basis of artistic skill. Suggest the following categories:

I. Photography
   - A. Underwater
   - B. Abovewater
   - C. Instamatic

II. Paintings and Drawings
   - A. Mood
   - B. Nature
   - C. People and the Sea

III. Sculpture

IV. Shell figures

V. Weaving (Coconut palm, Spartina grass, etc.)

VI. Jewelry

VII. Indian Art (Shell pendants, sharkbone necklaces, etc. See historical perspectives)

VIII. Dramatic presentations

IX. Anthology of Poetry or Short Stories

X. An original song

BIO-MOUNTING FISH AND OTHER MARINE SPECIMENS

INTRODUCTION

The state-of-the-art in environmental appreciation is to the point of look, but do not disturb. An exception can be made for very small numbers of specimens to be permanently mounted for examination by all of the students or when the animal or plant could not be returned to it's original condition because of accidental damage.

TO THE TEACHER

Often a specimen preserved in formalin or alcohol and stored in glass containers is not suitable for classroom use due to the fragility of glass and mess of formalin. An alternate method to display your specimens is to cast them in a polyethylene resin similar to fiberglass resin. Many commercially available mixes are obtainable from local hobby shops or biological supply houses as clear casting resin.

MATERIALS

Cupcake or pie tin
Cast-o-Lite (equivalent) casting resin
Hardener (comes with resin)
Parting agent (or car wax)
Sharp pointed object (pin, dissecting probe or better yet a glass tube drawn to a point)
Lint-free cloth
Acetone
Jewelers rouge
FAA—(50 cc denatured ethanol, 10cc formalin, 2cc glacial acetic acid, and 40 cc water)
Formalin solution (1 part to 9 parts water)

PROCEDURE

While algae, plants and soft bodied animals (worms, slugs, etc.) do not need preservation before casting, most soft-bodied organisms do need fixing in FAA for 12 hours. Most marine animals should be preserved in a 10% solution of formaldehyde for 24 hours and specimens whose body is more than 1cm thick should be injected to insure that bacterial action does not destroy the specimen after it is cast. The specimen should then be placed into methanol (undiluted) for 12 hours. Then gently place hard-bodied specimens on paper towels to drain. Transfer it to the resin (no catalyst) and soak for another 12 hours so that the plant or animal is an integral part of the finished block. Now your specimen is ready to be placed into a casting tin or mold of fresh resin with catalyst (hardener).

Coat the mold with a parting agent (usually available with the casting plastic) or use paste wax. Pour the mold (cupcake or small pie tin without ridges or creases) half full or less and let harden. This will provide a base for the specimen. Make sure that there is enough remaining depth for your specimen to be completely submerged in casting resin.

Gently lower the specimen into the mold and pour resin until the specimen floats or is partically covered. Work all the bubbles out of the resin with a sharp pointed object to insure clarity. Allow this resin to set and pour a third layer over the specimen, filling the mold completely. Remember to work the bubbles out again.

After a few hours your specimen will be ready to be removed from the mold. Do not take it out until the resin has completely hardened. It will shrink somewhat and come free of the mold itself. Failing this, place it in the freezer to help break the object free. Any cloudly or marked areas may be wiped clean with a lint-free cloth and acetone. A polishing with jeweler's rouge may help but in most cases is not necessary.

FRESH WATER AT THE SEA SHORE

MATERIALS

Spade or Shovel
Heavy clear plastic (3 meters x 3 meters)
Large mouth bottle or plastic frozen food container (1 liter)

PROCEDURE

Choose a sunny beach location where the tide will not wash in for several hours. Dig a hole in the sand about 1 meter across and 30 to 40 centimeters deep. Place the container at the center of the bottom of the hole. Cover the hole with the heavy clear plastic so that the plastic extends well beyond the edges of the hole in all directions. Anchor the plastic with heavy rocks (driftwood/boards) in such a way that allows the plastic to sag into the hole but does not touch the sides or bottom. Place a rock in the center of the plastic over the container. Allow the process to operate for several hours, or longer. Remove plastic carefully and examine the underside. Recover the container — is there a liquid in it? Taste it.

QUESTIONS

1. Is the moisture on the underside of the plastic salty?
2. What is the liquid in the container? Does it have any taste?
3. Explain the process that has taken place.
4. Does it have any commercial/industrial application?

Adapted from: Dept. of Commerce. Coastal Awareness, A Resource Guide . . . NOAA (pamphlet), G.P.O., Washington, D.C., 1978. 72 p.

HOW TO MAKE AN UNDERSEA THERMOMETER

MATERIALS

1. Standard laboratory thermometer 0°-100°C.
2. 2' piece of schedule 60 - 3" dia. PVC pipe (this is the PVC for electrical work not sprinkler systems. It withstands pressure better)
3. Cork 3" in diameter or greater
4. 2 dog teething rings with 3" inner diameter (optional)
5. Hack saw and drill
6. Cement mix
7. 3" PVC caps
8. Cable wire or nylon rope
   (Many materials might be furnished by students families if you ask.)

INTRODUCTION

1. Cut out two windows on PCV pipe, leaving about 20cm of one end entire for a water chamber.
2. Seal end on water chamber side and pour in cement about 3-6 cm high.
3. Cut two pieces of cork so that they fit snuggly inside PVC pipe.
4. Drill holes in top and bottom corks allowing for the thermometer to be held securely. Drill two additional holes in the bottom cork allowing water to fill the bottom chamber.
5. Place apparatus together (Thermometers should be removed, chamber emptied and cleaned between readings.)
6. Drill 3 holes in top and attach wire.

OPERATION

1. Drop apparatus attached to line to desired depth. While dropping fast, no water enters chamber.
2. Let apparatus sit momentarily, filling with water.
3. Lift apparatus and read temperature.

AERIAL PHOTOS FOR FREE!
(and ideas for using them)

There are several local sources of aerial photos from which a teacher may obtain free or very inexpensive aerial photos or photo-maps. First, a list of possible sources of free aerials:

- Local tax assessor or property appraiser — generally updates local photography every second or third year, *throws out* old photos. Scale 1"=200' or 1"=400'. May also provide free or inexpensive blueline copies of current photomaps.
- Local office of Soil Conservation Service (U.S. Dept. of Agriculture) — Can get aerials to pre-World War II, usually free, scale varies. Check them for Forest Service aerials too.
- Local Board of Realtors, Chamber of Commerce. Usually have good oblique aerials, may have some historical photos.
- Local Historical Society.
- Area Planning Board, County or City, Planning Dept. — A wealth of all kinds of photos, maps, charts, graphs, etc. Usually a planner around looking for an audience — use him!
- Local large industry. Check with public relations office. Particularly utility companies.
- Division of State Planning.
- State Department of Transportation — A wealth of finely detailed aerials, generally free.

IDEAS FOR USING AERIAL PHOTOS

Talk to a planner or an architect about this on an informal level. These two people especially have a wealth of ideas you can use in a classroom setting.

Corners: The more corners on a house, the more expensive it is, and the greater its energy comsumption level. Compare waterfront houses to low income houses Good statistical study. Check data with Property Appraiser.

Beaches: Photos of the same stretch of beach over the years show patterns of deposition, erosion. Make transparent overlay-maps. Pay special attention to inlets and passes.

My house: Often students can locate their own house on photos, especially those from property appraiser's office.

Habitat change: Document change from pinewoods to golf condo, etc. Acres lost, number of foxes lost, etc.

Growth of pavement: Alternatively pavement/unpaved ratio. Obviously, lots of room for math/statistical games in aerial photo-maps.

Satellite/High Altitude photos: EROS Data Center, Sioux Falls, S.D. 57201. A wealth of inexpensive maps, prints, slides. Write for catalog first! In Florida, DOT's Tallahassee office has satellite photos on a viewer, open to you on request.

Computer enchancement: Center for Wetlands, University of Florida is doing work on land use planning using computer-enhanced satellite photos. Check also Dept. of Geography at your local university or college.

Find a pilot: Check your local airport, ask for a pilot who will share expenses so you can take your own aerials.

Tips: Make sure windows are cleaned by pilot. Ask if window can be raised. Avoid shadow-images from glare on window.

Use UV or haze-cutter filter plus polaroid filter for best results.

Use SLR camera.

Don't touch side of plane when shooting

Plan flight ahead of time with pilot to avoid excess expense.

See section on photography.

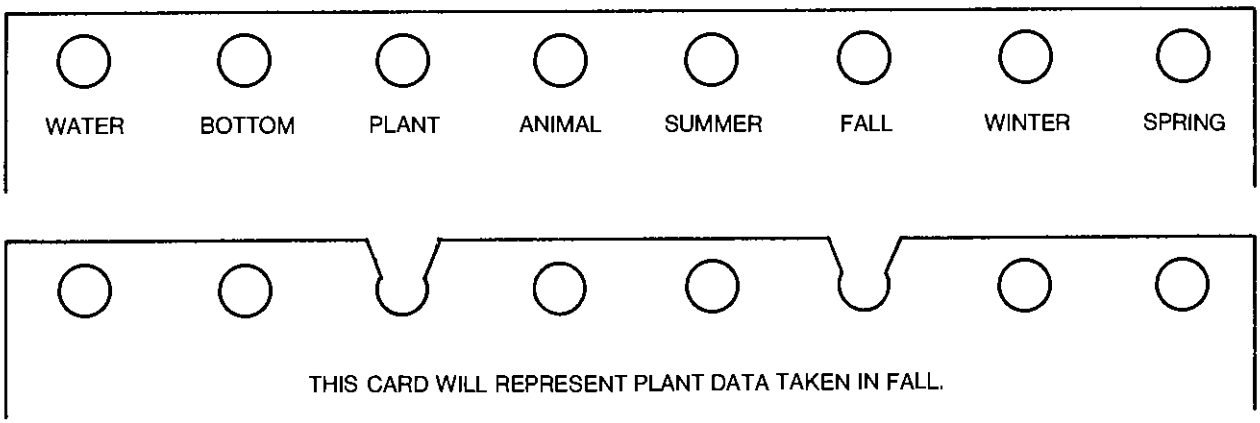
DATA FILE SYSTEM

Have a card punched along the edge with each hole representing a feature for data collecting. Holes may be assigned any value you wish to key on. This card will be the master card and is kept in the front of the file.

Each time a card is entered into the file, it is punched to represent the proper data.

By running a rod through the third hole and one through the sixth hole at the same time, all plant data taken in the fall can be drawn out.

Although each situation may require variations of the method, this system can be very useful in comparing and analyzing data over a period of time.



DATA FILE SYSTEM
SAMPLE OF POSSIBLE REPORT FORM

Front

Name of Body of Water _____ Date: _____

_____ Collected by _____

Meteorlogical Conditions

Barometer _____ Water Temperature _____

Thermometer _____ Water Turbidity _____

Wind Direction _____ Rainfall on date _____

Wind Speed _____ Other _____

Time of Sampling _____

Station No _____ Location _____

Land Marks _____ Bearing _____

_____ Bearing _____

Back

Depth _____ General description and/or sketch

Type of Bottom _____

Type of Sample _____

water, bottom, plant, fish _____

Physical size _____

_____

Color _____ Comments _____

_____ _____

Other (specify) _____ _____

_____ _____

_____

For ease of filing standard 5x8 card printed and punched McBee style

A QUICK AND EASY METHOD FOR PROCESSING LARGE AMOUNTS OF BRINE SHRIMP

(See Diagram page 158)

Some programs need large amounts of freshly hatched brine shrimp to feed corals and filter feeders in aquariums.

The set-up explained below uses materials found around every lab. With this equipment you should always have large amounts of eggshell-free shrimp to feed your animals. Students can quickly process large numbers of shrimp in only a few minutes each day.

EQUIPMENT NEEDED:

1. Three one-gallon cider jugs (more or less depending on needs)
2. One air pump
3. One 3-gang valve
4. Three air stones
5. One length of 6mm glass tube (two feet will do).
6. One piece of 3mm glass tube (4 inches).
7. About eight feet of air tube.
8. One #9 two-hole stopper.
9. One fine mesh filter net.
10. A short length of rubber tube.

EQUIPMENT SET UP AND MODIFICATION

1. Paint your cider jugs with opaque paint leaving a 5cm x 5cm window near the bottom of the jug.

2. Construct your siphon apparatus as follows:
   a) Bend the glass tube as shown in diagram #1.
   b) Slide the stopper up the tube and adjust so that the rubber tip you will slip on the lower end just touches the bottom of the jug.
   c) Slide the small diameter tube into the second hole.

3. Diagram #2 illustrates one layout of the equipment. Make your layout according to your own specifications.

4. Brine solution: use ten teaspoons of non-iodized table salt per gallon of water. To this add two drops of Chlorine remover.

5. Funnel 1-2 quarts of brine into your jugs. Add up to one teaspoon of brine shrimp eggs to one jug. Drop in an air stone and in approximately 48 hours, your eggs will hatch. Test for hatching by placing a light near the jug "window." Shrimp will migrate toward the light.

6. After your eggs have hatched, remove the air stone and put in your siphon apparatus. Place the jug on a raised base, tilt the jug forward with a block of wood placed opposite the light opening. The eggshells will float to the top. The shrimp will concentrate around the light opening where the lower end of your siphon tube is. After a few minutes, place a container under your outside siphon tube. Start your siphon by blowing on the small tube in the stopper. Stop the siphon just before all the water runs out, this prevents the floating eggshells from getting into the siphoned water.

7. Pour the siphoned solution through your filter net to remove the shrimp. Rinse the shrimp in fresh water and they are ready for use. You can use the brine solution again if you wish.

8. By starting eggs hatching one day apart in three jugs, you can be assured of a hatch each day. Just be sure you keep track of your hatching order.

INVERTEBRATE AQUARIUM
(under 50 gallons)

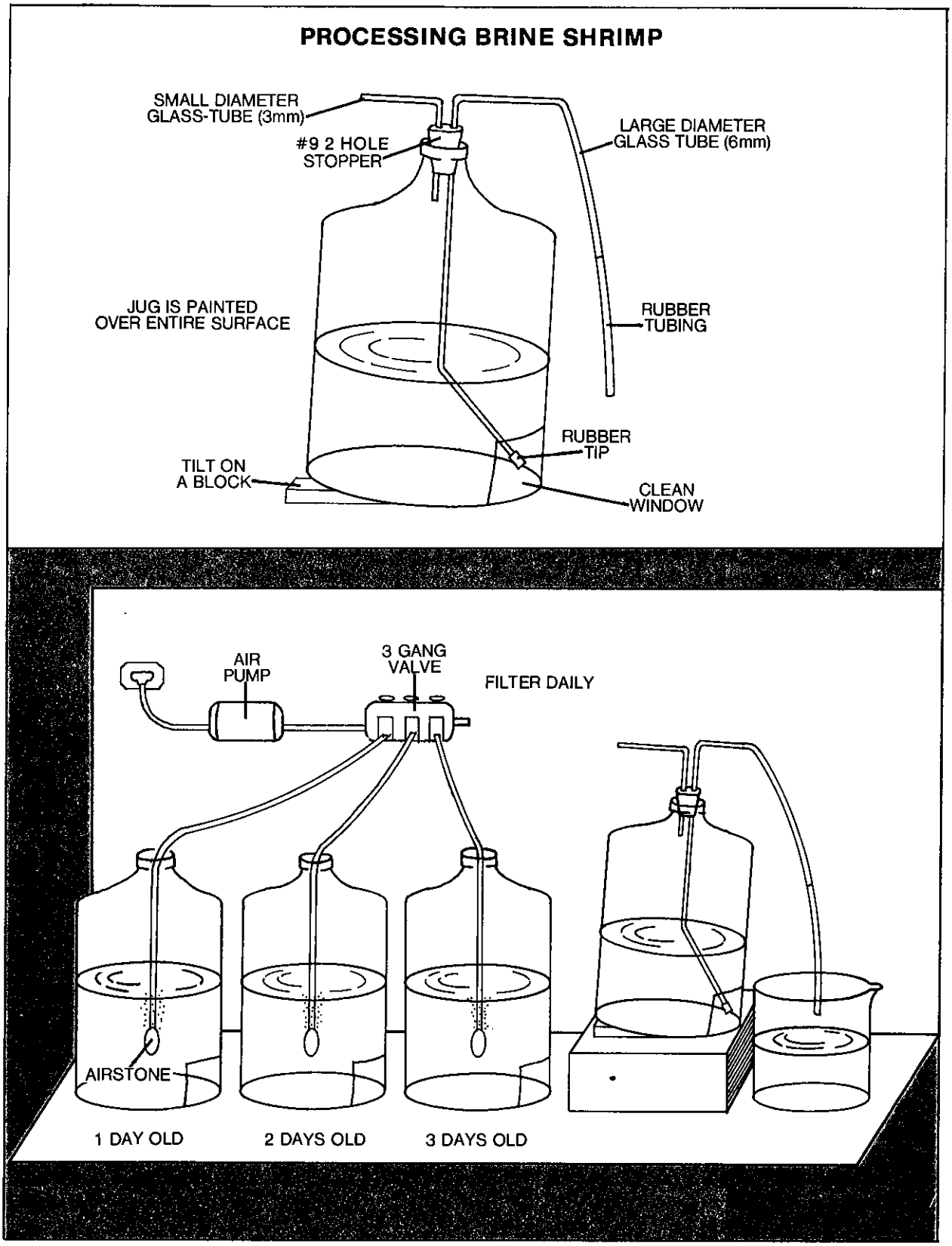
NEEDED

1. All-glass aquarium.
2. Bottom filter or plastic egg-crate covered by fiberglass screen with plastic tube for air release.
3. 12" of crushed coral (preferably), silica sand, or washed shell (approximately 1½ pounds per gallon of water) over bottom filter.
4. Invertebrates (approximately 1" per gallon of water) Note: Salt water fish (vertebrates) are more active than fresh-water fish and salt water invertebrates. An aquarium should contain no more than 1 fish per 2 gallons of water. Therefore if an aquarium contains 2 fish that are an inch long, it can contain only 6 inches of invertebrates with it.
5. Pump and hose.
6. Salt water.

PREPARATION

1. Place bottom filter and cover with crushed coral. Beach sand is too fine in size. The crushed coral serves several functions. Animal waste makes the water too acid. The crushed coral is alkaline and neutralizes this acid effect. Secondly the crushed coral provides a home for aerobic bacteria (when air bubbles through it) which decompose the organic wastes.
2. Pour in clear salt water from the ocean. Any cloudiness should clear in 24 hours.
3. Put in a dead fish or invertebrate for a day or two and then remove. This builds up bacteria in your under-gravel filter. These bacteria will take organic matter out of the water and deposit the decomposed results under the gravel keeping your water clean.
4. Allow aquarium to "age" at least one week before adding invertebrates. If coelenterates are added, in particular corals, bag them under water and do *not* expose them to air. Corals needs a "Gro-light" for the algae living in them. Scallops and corals need plankton to eat. (See tips on raising brine shrimp.)
5. You will want to change 10% of the water every 2 or 3 weeks. Add *fresh* water to compensate for evaporation loss. A dust cover will reduce evaporation loss.
6. Make a list of observations and information you have gained through keeping an aquarium.

PROCESSING BRINE SHRIMP



COLLECTING

In order to collect in Florida's marine environment, one must obtain a permit issued by the Department of Natural Resources, Division of Marine Resources, Tallahassee, FL 32304.* This permit enables the scientific instructor to collect marine organisms, except endangered species, provided the collecting is not done within an aquatic preserve, a Florida State Park or any of the federal refuges or sanctuaries. In most instances, the permit is restricted to certain counties. Be certain that you contact the local office of the Marine Patrol, an agency of the Florida Department of Natural Resources.

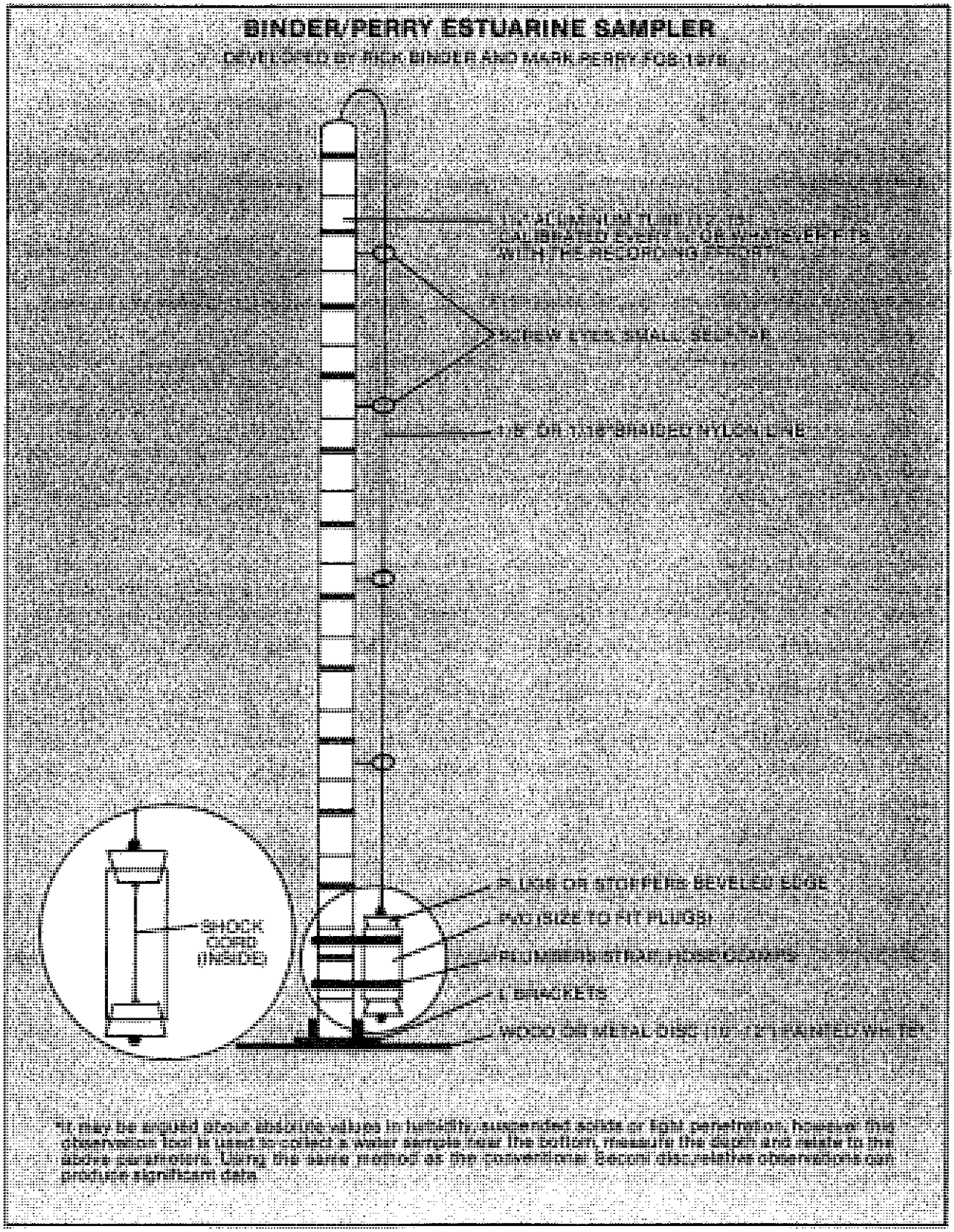
Once in the field, the students and teacher must take care to avoid overcollecting and the total disruption of the ecosystem. If rocks are overturned, be certain to return them to their original position. Remember that some populations are slow to recover once overcollected. A good example of this is the scarcity of Queen Conchs in the Florida Keys, an area in which they were formerly abundant.

Throughout field work a conservation ethic must be presented to the participants. Without this attitude the reason for field trips will be lost.

COASTAL CONSERVATION

1. Replace each organism as it was found.
2. Leave nothing behind except footprints.
3. Avoid over-structuring; relax and take advantage of the events as they happen.
4. Encourage everyone to look closely, take time to see what is going on.
5. As organisms are discovered, discuss them, holding them for all to see, and allowing anyone to handle them who cares to. Replace them when finished.

**Other states may also require a collecting permit.*



*It may be argued about dissolved oxygen in turbidity, suspended solids or light penetration, however this observation tool is used to collect a water sample near the bottom, measure the depth and relate to the above parameters. Using the same method as the conventional, secchi disc relative observations can produce significant data.

SELECTED REFERENCES AND FILM SOURCES

Identification of organisms whether unusual or common is sometimes a difficult task. The search for specific taxonomic "keys" with good illustrations and enough, but not too much, technical presentation can be at times frustrating. The References listed here include guides and other sources which the authors found through their experience to be very useful.

Periodicals, newsletters and magazines have not been included here because of their nature as every-growing or otherwise changing sources of information. These publications, however, present some of the most updated material in the field, and it recommended that the individual review them for their particular preference.

The Film Sources section gives a selection of some commonly used sources for educational films. Each has their own catalogue and it is suggested that from each catalog a film list for marine science topics can be developed.

AQUATIC ORGANISMS

FRESHWATER

A Guide to the Study of Freshwater Biology by Paul R. Needham and James G. Needham.

Keys and descriptions to aid recognition of freshwater algae, invertebrates, and fishes. Line drawing illustrations. 108 pages. Paperback. 1962. Published by Holden-Day Inc., 500 Sansome Street, San Francisco, California 94111.

Florida Fishes by Rube Allyn

Not complete, but good for openers, including fresh and salt-water fishes. Black and white plus a few color illustrations. 90 pages. Paperback. Published by the Great Outdoors Publishing Company, 4747 28th Street North, St. Petersburg, Florida 33714. 1969.

Guide to the Reptiles, Amphibians, and Fresh-Water Fishes of Florida by Archie Carr and Coleman J. Goin.

Good coverage of Florida fishes and herps. Includes keys and instructions concerning their use. Some black and white illustrations. 341 pages. Hardback. Published by U. of Fla. Press, Gainesville. 1959

MARINE

A Handbook for Beach Strollers by Donald J. Zinn.

A readable, entertaining little book describing the ecology of seaside creatures. It was written for the northeast coast, but many of the same genera can be found off the coast here. Available from Marine Advisory Service, University of Rhode Island, Narragansett Bay Campus, Narragansett, Rhode Island 02882. 1974.

Atlantic Reef Corals by F.G. Walton Smith.

A handbook of the common reef and shallow-water corals of Bermuda, Florida, the West Indies, and Brazil. 112 pages. Hardback. Black and white photographs. Published by University of Miami Press in 1948. Recently revised.

Field Book of Seashore Life by Roy Waldo Miner.

A compact manual of more than 1300 common invertebrate marine animals found in the shallow waters of the North Atlantic Coast. Most invertebrates found off the coast of Southern Florida can be identified to family using this book, although genera and species are usually different. Line drawing illustration. 888 pages. Hardback. Published by G.P. Putnam's Sons, New York, 1950.

Seashells of North America by R. Tucker Abbott.

A good, reasonably complete, easily useable guide for shell identification. Many color illustrations. 280 pages. Paperback. Published by Golden Press, New York. 1968.

Seashores by Herbert S. Zim and Lester Ingle.

A golden Nature Guide to seashore plants and animals. Color illustrations. 160 pages. Paperback. Published by Golden Press, New York. 1955.

Shelling and Beachcombing in Southern and Caribbean Waters by Gary M. Dukane.

Common shells make up much of the book, and Abbott (above) is better for identification. But the section on other flotsam and jetsam including "Sea Beans," makes the book a must for beachwalkers. Good illustrations. Published by Dukane Press, Inc., 2901 Simma Street. Hollywood, Florida 33020.

Caribbean Reef Fishes by John E. Randall.

Formal accounts are given of the 300 species most likely to be observed by man in the sea or caught by man near shore. Mention is made of more than 100 other fishes, usually with sufficient information to permit their identification as well. The 300 species are all illustrated more than half in color. 318 pages. Hardback. Distributed in the U.S.A. by T.F. H. Publications Inc., 211 West Sylvania Avenue, P.O. Box 27, Neptune City, N.J. 07753. 1968.

Field Book of Marine Fishes of the Atlantic Coast by Charles M. Breder.

Line drawings, keys, economic and ecological role of salt-water fishes. Although old, with keys that are sometimes hard to use, the line drawings and the

coverage of estuarine and near-shore species make it a valuable guide. G.P. Putnam's Sons. 1948.

Fishes of the Bahamas and Adjacent Tropical Waters by James E. Bohlke and Charles C.G. Chaplin.

A monumental, comprehensive, beautifully illustrated work. (Also expensive, of course.) 771 pages. Hardback. Published by Livingston Publishing Company, Wynnewood, Pa. 1968, for the Academy of Natural Sciences of Philadelphia.

Fishwatchers Guide to West Atlantic Coral Reefs by Charles C.G. Chaplin.

A drastically shortened version of the book listed above designed for use in the field, which in this case means in the water. Consequently the entire book is printed on polyolefin which resists water damage. Color illustrations. Polyolefin-back! Published by Livingston Publishing Company, Wynnewood, Pa. 1972.

The Sharks Around Us by Dr. R.D. Skocik

A complete study of sharks found in Florida waters including identification (including teeth), morphology, shark attach information and shark fishing. Published by Stan Publishing Co., Inc., 609 N. Railroad Avenue, Boynton Beach, Florida 33435. 1971. Available in local bookstores. Paperback.

Seashore Life of Florida and the Caribbean by Gilbert L. Voss.

Color pictures and line drawings of common marine invertebrates. 168 pages. Hardback. Published by E.A. Seemann Publishing, Inc., Miami, FL.

Tropical Marine Invertebrates of Southern Florida and the Bahama Island by Warren Zeiller, 1974, Wiley, N.Y.

Color photos, some description. This plus Voss covers the field admirably.

The Oxford Book of Invertebrates by Nichols, David and John Cooke and Derek Whiteley, 1971, Oxford, London.

Includes terrestrial, fresh and salt water organisms with descriptions and color plates.

Guide to Corals and Fishes of Florida, the Bahamas and the Caribbean by Idaz Greenberg, 1977, Seahawk, Miami.

High quality color drawings plus descriptive information of 260 species of fish and corals, including gorgonians. Paper.

MARINE, GENERAL

Life In and Around the Salt Marshes by M.J. Ursin, 1972, Crowell, N.Y.

Field Guide to Some Carbonate Rock Environments of the Florida Keys and Western Bahamas by H. Gray Multer, 1971, Fiarleigh Dickinson University, Madison 17940.

Land From the Sea by John E. Hoffmeister, 1974, U. of Miami, Coral Gables.

The Sea Brings Forth by Jack Rudloe, 1968, Knopf, N.Y.

Entertainingly accurate, readable natural history of benthic organisms recovered from shrimp trawls.

The Erotic Ocean by Jack Rudloe, 1970.

The Encyclopedia of Oceanography by Rhodes W. Fairbridge, 1966, Reinhold, N.Y. Academic.

Waves and Beaches by Willard Bascom, 1964, Doubleday, N.Y.

The dynamics of the ocean surface, readable and accurate. The basic descriptive text, with diagrams and photos. Paper.

DANGEROUS MARINE ORGANISMS

Dangerous Sea Creatures by Thomas Helm, 1976, Funk and Wagnalls, N.Y.

Photos, drawings and descriptive text for venomous, poisonous and otherwise dangerous vertebrates and invertebrates.

Atlas of Aquatic Dermatology by Alexander Fisher, 1978, Harcourt, N.Y.

A medical text which includes color photos and treatment of stings caused by marine organisms. Your doctor may be able to get a copy free from Lederle Laboratories. Paper.

The Sting of the Sea by Florida Dept. of Natural Resources, 1978.

A one-page handout on organisms and very basic first aid.

How to Cope with Dangerous Sea Life by Iversen/Skinner, 1976, Windward, Miami.

NATIVE PLANTS

A Flora of Tropical Florida by Robert Long and Olga Lakela.

Comprehensive coverage of the plants in this area, but difficult to use unless you have a good botanical vocabulary and some experience in using keys. 962 pages, including relatively few line drawing illustrations. Hardback. Published by University of Miami Press, 1971.

A Guide to Common Florida Salt Marsh and Mangrove Vegetation by Jedfrey M. Carlton.

A 30 page booklet describing common salt marsh plants. Includes black and white photographs. Published by Florida Department of Natural Resources Marine Research Laboratory, 1975. Available from them at 100 Eighth Ave., S.E., St. Petersburg, Florida 33701.

The Trees of South Florida by Frank Craighead, 1971, U. of Miami, Press.

Details the plant communities of extreme southern Florida, with an excellent treatment of coastal systems.

Everglades Wildguide by Jean Craighead George, 1972, Nat. Park Serv. U.S. Dept. Interior.

Distinctive color drawings and diagrams outline the systems, communities and major species of the Everglades. Paper.

The Native Trees of Florida by Erdman West and Lillian Arnold, 1956, University of Florida, Gainesville.

A descriptive key with line drawings. Paper.

Aquatic Weed Identification and Control Manual by Alva P. Burkhalter and others.

Unfortunately this booklet treats virtually every common aquatic plant as a weed to be controlled. Nevertheless, the color pictures and detailed descriptions are a great help in identification too. Published by the Bureau of Aquatic Plant Research and Control, Department of Natural Resources, Larson Building, Tallahassee, Florida 32304.

Guide to Plants of the Everglades National Park by Alex D. Hawkes.

A booklet that outlines the major plant families found within the park and discusses some of the more commonly encountered members of each family. Locations are given to help you find the plants described. 50 pages with some black and white photographs. Paperback. Published by Tropic Isles Publishers, Inc., P.O. Box 613, Coral Gables, Florida 33134. 1965. Available from Everglades Natural History Association Inc., P.O. Box 279, Homestead, Florida 33030.

Trees of the Everglades National Park and the Florida Keys by George B. Stevenson.

A compact, easy-to-use guide. Trees are grouped according to obvious characteristics, and each tree included has a line drawing illustration. 32 pages, Paperback. Published by the author in cooperation with the Everglades Natural History Association Inc., P.O. Box 279, Homestead, Florida in 1969.

Wild Plants for Survival in South Florida by Julia F. Morton.

Descriptions of over 100 edible native plants with photographs in black-and-white and color. Paperback with 80 pages. 1968. Published by Hurricane House, Inc., 14301 SW 87th Ave., Miami, Florida 33158.

Wildflowers of the Southeastern United States by Wilber H. Duncan and Leonard E. Foote.

Although the southern tip of Florida is specifically excluded from the scope of this book, many of the plants found here are incidentally included. Very tropical species that do not occur in other parts of the Southeast are left out. The book is included on this list because it is recent and the color photographs make it easier to use than the other wildflower book listed. 296 pages. Hardback. Published by University of Georgia Press, Athens, Georgia, 1975.

BIRDS

A Field Guide to the Birds by Roger Tory Peterson

The classic field guide to birds, now in its 45th printing, with several revisions along the way. Color illustrations. 230 pages. Hardback and paperback. Published by Houghton Mifflin Company, Boston. 1947.

Birds of North America by Chandler S. Robbins, Bertel Brunn, & Herbert S. Zim.

Another excellent field guide to birds, including distribution maps and color illustrations. 340 pages. Paperback. Published by Golden Press, New York. 1966.

REPTILES AND AMPHIBIANS

A Field Guide to Reptiles and Amphibians by Roger Conant

Part of the Peterson Field Guide Series. Good, useable field guide with many color and some black and white illustrations. Includes distribution maps. 366 pages. Recently revised edition. Hardback and Paperback is now available. Published by the Riverside Press, Cambridge Mass. 1958.

Snakes of Florida by Owne Godwin

Small (48 pages) compact paperback limited to Florida snakes, but including good color pictures and fairly detailed information. Published by Godwin's Gatorland, U.S. 17-92-441, Kissimmee, Florida 32741. 1973.

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U.S. Navy Hydrographic Office, *American Practical Navigator,* H.O. Pub. No. 9, G.P.O., Washington, D.C., 1962, p. 1524.
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FILM SOURCES

Encyclopedia Britanica Educational Corp.
425 N. Michigan Avenue
Chicago, Illinois 60611
(312) 321-6800

Modern Talking Picture Service, Inc.
2323 New Hyde Park Road
New Hyde Park, New York 11040

McGraw-Hill Films
McGraw-Hill Book Company
110 15th Street
Del Mar, California 92014
(714) 453-5000

Cornet Instructional Media
65 East South Water Street
Chicago, Illinois 60601
(312) 977-4000

Moody Institute of Science
12000 E. Washington Blvd.
Whittier, California 90606
(213) 698-8256

Motion Picture Service
NOAA-U.S. Department of Commerce
12231 Wilkins Avenue
Rockville, Maryland 20852
(301) 443-8411

Landsat Educational Film Series
NASA Kennedy Space Center
Public Affairs Office, Code PA-EPB
Kennedy Space Center, FL 32899

Commanding Officer
Naval Education and Training
Support Center
Atlantic Naval Station
Bldg., Z-86
Norfolk, VA 23511

Branch of Visual Services
U.S. Geological Survey
303 National Center
Reston, VA 11040
(516) 437-6300

TABLE OF CONVERSION FACTORS*

(U.S. units to metric units)

Length

1 inch	= 25.4 mm.
	= 2.54 cm.
1 foot	= 30.48 cm.
	= 0.3048 m.
1 statute mile	= 1.609 km
1 nautical mile	= 1.853 km.

Area

1 sq. in.	= 6.45 $cm.^2$
1 sq. ft.	= 929.03 $cm.^2$
	= 0.0929 $m.^2$

Volume and capacity

1 cubic inch	= 16.39 cc.
1 cubic foot	= 28,317 cc.
	= 26.317 liters
	= 0.028317 cu. m.
1 quart	= 0.946 liter

Weight

1 ounce	= 28.35 gm.
1 pound	= 453.6 gm.
	= 0.454 kg.
1 short ton	= 907.2 kg.

Pressure

1 p.s.i.	= 70.3 $gm./cm.^2$
	= 0.0703 $kg./cm.^2$
	= 0.703 meter of fresh water
	= 5.17 cm. Hg.
1 in. of fresh water	= 25.4 mm. water
	= 2.54 $gm./cm.^2$
1 in. of mercury	= 25.4 mm. Hg.
	= 34.54 $gm./cm.^2$

TABLE OF CONVERSION FACTORS*

(Metric units to U.S. units)

Length

1 cm.	= 0.394 in.
1 meter	= 39.37 in.
	= 3.28 ft.
1 kilometer	= 0.621 mi.

Area

1 $cm.^2$	= 0.155 sq. in.
1 $m.^2$	= 10.76 sq. ft.
1 sq. km.	= 0.386 sq. mi.

Volume and capacity

1 cc. or ml.	= 0.061 cu. in.
1 cu. m.	= 35.31 cu. ft.
1 liter	= 61.02 cu. in.
	= 0.035 cu. ft.
	= 33.81 fl. oz.
	= 1.057 quarts

Weight

1 gram	= 0.035 oz.
1 kg.	= 35.27 oz.
	= 2.205 lb.

Pressure

1 $gm./cm.^2$	= 0.394 inch of fresh water
1 $kg./cm.^2$	= 14.22 p.s.i.
	= 32.8 feet of fresh water
	= 28.96 inches of mercury
1 cm. Hg.	= 0.193 p.s.i.
	= 0.446 foot of fresh water
	= 0.394 inch of mercury
1 cm. of fresh water	= 0.394 inch of fresh water

*Data from U.S. Government Printing Office, 1963, U.S. Navy Diving Manual. Part I. Government Printing Office: Washington, D.C.

TEMPERATURE CONVERSION FORMULAE

Fahrenheit to Celsius — $C° = 5/9\ (F°-32)$

Celsius to Fahrenheit — $F° = 9/5\ (C°+32)$

TABLE OF CONVERSION FACTORS*

(Metric units to other metric units)

Length

1 millimeter (mm.)	= 0.1 cm.
	= 0.001 m.
1 centimeter (cm.)	= 10 mm.
	= 0.01 m.
1 decimeter (dm)	= 100 mm.
	= 10 cm.
	= 0.1 m.
1 meter (m.)	= 1000 mm.
	= 100 cm.
	= 10 dm.
	= 0.001 km.
1 kilometer (km.)	= 1000 m.

Volume and Capacity

1 cubic centimeter (cc.) (or 1 millimeter (ml.))	= 0.001 liter
1 liter (l.)	= 1000.027 cc.
	= 1000 ml.
	= 0.001 cu. m. ($m.^3$)
1 cubic meter ($m.^3$)	= 100 l.

General System of Multiples

Multiple	Prefix	Symbol
10^{12}	tera	T
10^{9}	giga	G
10^{6}	mega	M
10^{3}	kilo	k
10^{2}	hecto	h
10	deka	da
10^{-1}	deci	d
10^{-2}	centi	c
10^{-3}	milli	m
10^{-6}	micro	μ
10^{-9}	nano	n
10^{-12}	pico	p
10^{-15}	femto	f
10^{-18}	atto	a

Area

1 sq. cm. ($cm.^2$)	= 100 $mm.^2$
1 sq. m. ($m.^2$)	= 10,000 $cm.^2$
1 sq. km. (km^2)	= 1,000,000 $m.^2$

NOTE—European usage employs a comma where we use a decimal point and a period where we use a comma (in large numbers).

Weight

1 milligram (mgm.)	= 0.001 gm.
1 gram (gm.)	= 1000 mgm.
	= 0.001 kg.
1 kilogram (kg.)	= 1000 gm.

Weights of fresh water

1 cc. or 1 ml.	= 1 gm.
1 liter	= 1 kilogram

Pressure

1 gram per square centimeter ($gm./cm.^2$)	= 0.001 $kg./cm.^2$
	= 1 cm. of fresh water
1 kilogram per square centimeter ($kg./cm.^2$)	= 1000 $gm./cm.^2$
	= 10 meters of fresh water
	= 9.75 meters of sea water
	= 73.56 cm. Hg.
	= 0.968 atm.
1 centimeter of mercury (cm. Hg.)	= 13.6 $gm./cm.^2$
	= 13.6 cm. of fresh water
1 centimeter of fresh water	= 1 $gm./cm.^2$
1 atmosphere	= 1.033 $kg./cm.^2$
	= 760 mm. Hg.

*Data from U.S. Government Printing Office, 1963, U.S. Navy Diving Manual. Part I. Government Printing Office: Washington, D.C.

TABLE OF CONVERSION FACTORS*

(U.S. units to other U.S. units)

Length

1 inch (in.)	= 0.083 ft.
1 foot (ft.)	= 12 in.
1 yard (yd.)	= 36 in.
	= 3 ft.
1 fathom	= 6 feet
1 statute mile	= 5,280 feet
1 nautical mile	= 6,080 feet
	= 2,026.7 yd.

Area

1 sq. in.	= 0.069 sq. ft.
1 sq. ft.	= 144 sq. in.
1 sq. yd.	= 1,296 sq. in.
	= 9 sq. ft.
1 acre	= 43,560 sq. ft.
	= 0.00156 sq. mi.
1 sq. mile	= 640 acres

Volume (cubic measurements)

1 cu. in.	= 0.00058 cu. ft.
1 cu. ft.	= 1,728 cu. in.
	= 29.92 quarts
	= 7.48 gallons
1 cu. yd.	= 27 cu. ft.

Capacity (liquid measure)

1 pint (pt.)	= 18 fluid ounces
	= 28.88 cu. in.
1 quart (qt.)	= 2 pt.
	= 57.75 cu. in.
1 gallon (gal.)	= 4 qt.
	= 231 cu. in.

Weight (avoirdupois)

1 ounce (oz.)	= 0.0625 lb.
1 pound (lb.)	= 16 oz.
1 short ton	= 2,000 lb.

Weights of water

1 quart	= 2 pounds (fresh water)
1 cu. ft.	= 62.4 lbs. (fresh water)
	= 64 lbs. (sea water)

Pressure

1 pound per square inch (p.s.i.)	= 2.31 feet of fresh water
	= 2.25 feet of sea water
	= 0.068 atm.
	= 2.036 in. Hg.
1 atmosphere (atm.)	= 14.696 p.s.i.
	= 29.92 in. Hg.
	= 33.9 ft. of fresh water
	= 33 ft. of sea water
1 foot of sea water	= 0.445 p.s.i.
1 inch of mercury (in. Hg.)	= 0.491 p.s.i.
	= 1.133 feet of fresh water
	= 13.60 inches of fresh water

*Data from U.S. Government Printing Office, 1963, U.S. Navy Diving Manual. Part I. Government Printing Office: Washington, D.C.

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