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Division of Marine Sciences
(TREDMAR Programme)

FIELD WORK IN MARINE ECOLOGY FOR SECONDARY SCHOOLS IN TROPICAL COUNTRIES

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INTRODUCTION

Many schools in the tropics are close to coastal marine environments. The oceans contain many forms of life that are of great interest in biology. Life first evolved in the sea, and many primitive types of plants and animals occur there. Any school within reach of the sea should take advantage of this great natural laboratory.

The intertidal zone along the shore that is alternately covered and uncovered by the tides is the easiest area to reach for class field activities. Almost every activity in this section can be done by pupils on the shore when the tide is out. A few can be extended into shallow water by students who are good swimmers. In areas with little tidal variation, it may be necessary to wade into shallow water to observe features of interest.

There are four major habitats in tropical intertidal areas: sandy-muddy areas, mangroves, coral reefs and rocky shores. Only one or two of these will be available to most schools, but enough activities are proposed to demonstrate basic biological concepts in any of these environments.

Sandy and muddy habitats have a substrate of fine sediment that is too mobile for many organisms to attach to, particularly in the wave zone. Seagrasses or seaweeds may root in the sand or attach to small rocks and shells, and many animals will live on or burrow in the sediment. It is often necessary to dig up the top layer of sand or mud to discover this interesting community.

Mangrove trees are specially adapted to growing in salty water rooted in anoxic sediment. They are to be found mainly in estuaries and muddy coastlines where freshwater and saltwater mix, and each species may have precise salinity requirements. Their roots help to trap sediment and create a protected environment, while providing a place for many algae and animals to attach and live. Mangrove leaves are the beginning of an important detrital food chain.

Coral reefs are major geological structures built by biological activity through the accumulation of the skeletons of corals and coralline algae and of sand made from algae and animal shells. The coral reef ecosystem is very complex, with more different species crowded into a small area than almost any other habitat on earth.

Rocky shores are less common in the tropics than in temperate areas. Their hard substrates provide attachment for many kinds of plants and animlas, which compete for space in the intertidal and high subtidal. Many man-made shorelines consist of concrete walls, large rocks or pilings which may develop similar communities to rocky shores when they have been in the sea long enough.

Life is not easy in the intertidal zone. While light is abundant and nutrients are often available, it is necessary to attach to something to keep from being swept away by the force of waves and currents. Space is often at a premium, leading to fierce competition. It is necessary to withstand extreme temperatures and alternate wetting and drying, as well as salinities from fresh rainwater to pure salt crystals. Growth can be rapid, but predators are numerous. The organisms of the intertidal show many different strategies for adapting to this extreme environment.

Class field work in the intertidal also presents some special problems and dangers. Where there is a small tidal range, it may be necessary to wade and to look at the bottom through the water's surface. Reflections and water movement can make this difficult, and pupils may need to use glass-bottomed buckets or boxes, facemasks or goggles to see clearly. Where the tidal range is great, the intertidal will be more accessible at low tide, but attention must be paid to the incoming tide which can rise quite rapidly. Obtain accurate tide data so that field work can be planned for a low tide period. Start either at low tide at the water's edge and work towards the shore, or follow the low tide out and in. Waves can also surprise unwary students, so care must be taken to choose conditions and sites with an adequate margin of safety. Students must always be supervised closely while in the field.

Some dangerous animals may be encountered in coastal waters as on land, so teachers should inform themselves about those which occur locally (cone shells, stone fish, etc.) and warn the pupils about them before going into the field. Many animals have sharp shells or edges, and care must be taken to avoid cuts and scrapes. Pupils must wear adequate footware such as shoes of rubber or canvas with soles of sufficient thickness to protect feet from cuts, spines and stings. Protection from the sun may also be advisable. A first aid kit should always be at hand just in case. With these simple precautions, a class field trip to the intertidal can be a safe and rewarding experience.

1. COLLECTIONS OF MARINE LIFE

(A; I, II)

Objectives

1. to observe marine plants and animals closely.
2. to prepare herbarium mounts and other speciments.
3. to identify organisms.
4. to understand the principles of classification.

Curriculum links

Identification and classification, species, biological diversity.

Preparation

Habitat:	any accessible coastline, preferably with a hard bottom and attached plants and animals.
Time required:	minimum 2 hours in the field, not counting the time to reach the coastal habitat, plus 2 - 3 hours in the classroom or laboratory to work on the collections.
Teacher:	obtain and study whatever reference works and keys are available locally for the identification of marine life. Illustrated works are better for student use. Decide what should be collected and what the collections will be used for.
Students:	the major groups of marine plants and animals should be studied before the field activity.
Equipment:	small knives for prying organisms off hard surfaces shovels or trowels for digging in sediments small plastic bags for collecting specimens small bottles or vials for fragile specimens buckets for carrying specimens and equipment small pieces of heavy paper for labels cards or heavy drawing paper for herbarium mounts pencils hammer and chisel (supervised by teacher) small dip nets (made from wire and mosquito screen) shallow trays or dishes for sorting old newspapers pieces of muslin to cover herbarium mounts aquaria or large glass jars hand lense microscope or dissecting microscope (optional).

Strategy

Question: What plants and animals live in the coastal marine environment?

1. Collecting organisms is an excellent way to learn to observe them closely. However, the amount of collecting by the class must be carefully controlled by the teacher, as a large class collecting too many organisms can have a serious effect on the environment. Organisms should not be collected except for a definite purpose.

2. Each pupil or small group of pupils is supplied with collecting tools (knife, shovel, dip net), bucket, plastic bags, bottles, labels or notebook, pencil.

3. Pupils should be instructed to make a collection of certain common organisms such as:

   algae and seagrasses
   small common molluscs or shells
   small crabs, crustacea, isopods.

4. In addition, one example of each plant and animal observed can be collected for the whole class. Duplicate specimens should be returned to where they were collected.

5. For each collection, a label should be made with the following information: name of collector, date, place name, where the organism was found, type of substrate or habitat, associated organisms, size of the whole organism (if only a part was taken), relation to tide level. The organism and label should be put together in a plastic bag or bottle. Alternatively, the specimens can be numbered and the information written down in a small field notebook. Space should be left under each entry to add the name and other data once identification is made.

6. Algae and most animals should be kept damp but not under water. Only swimming animals should be kept in water. Avoid overheating in the sun.

7. If possible, the class should return immediately to the school laboratory or other work area to sort and prepare the collections and to observe the living animals. If more than 1 day will pass before the collections can be worked on, they should be held indoors in large containers or aquaria of seawater or preserved in 3% formaldehyde in sea water. Preserved collections should be rinsed before being given to the students for sorting. Formaldehyde should not be used in containers or work areas which will serve for holding or rearing live organisms, as the fumes and residue are toxic to delicate organisms.

8. In the laboratory, collection should be sorted and identified, with the names and other information added to the labels or notebooks. If references giving the scientific names are not available, then common names or local names should be used.

9. If time permits, observe the special features, morphology and behaviour of the organisms collected. Place the organisms in trays or aquaria to make observation easier. Examination with a hand lense or microscope can also help students to see smaller organisms and features.

10. Specimens can be prepared for a permanent class collection. Algae are laid out or floated out on heavy paper or cards, then covered by a piece of muslin and dried between several sheets of newspaper in a plant press or under a heavy weight. The newspaper should be changed every day until the algae are dry. Interleaving with corrugated cardboard can speed drying. Shells are cleaned, and most other hard-bodied animals can be air-dried (outside, they will smell!). Soft-bodied animlas must be preserved in 70% alcohol.

11. Pupils can also make personal collections of the common algae, shells and crustacea they have collected.

12. The completed collections should be arranged according to the scientific classification of organisms, if that is available. The teacher should explain the distinguishing characteristics of the organisms. If a scientific classification is not possible, an artificial classification can be made as follows:

    - Decide on the criteria for separating the organisms (colour, shape, anatomy, etc.)

    - Separate and classify the organisms according to the criteria adopted.

    - Make any revisions in the criteria that seem necessary.

13. On the basis of the complete collection of all the plants and animals observed, make a species list for the area showing how many different kinds of organisms occur there.

Teaching hints

There should be no uncontrolled class collecting, as this can rapidly strip an environment. Too often animals collected by classes are not looked at again but are left to rot in various containers in the laboratory. This should be prevented. There should be a definite purpose in mind for any collection made. Restrict multiple student collections as much as possible to organisms that are very abundant and are known to regenerate quickly.

It is not necessary that the identifications and classifications made by the class always be scientifically accurate as this often requires reference to complex scientific works that are inappropriate at the second school level. It is the scientific methods of observation, identification and classification that should be understood. However, students should be encouraged to differentiate between forms at the species level.

Follow up

This activity is a good preparation for many of the ecological activities that follow, as it helps to ensure that different types of organisms are familiar to the students.

2. SANDY-MUDDY SHORE ZONATION

(A, B; I, II)

Objectives

1. to discover the organisms that live on or in sand and mud.
2. to observe the relationship between environmental factors or gradients and organism distributions.
3. to learn the principles of quantitative sampling.
4. to process and present numerical data and to test it for statistical significance.

Curriculum links

Identification of organisms, distributions, gradients of environmental factors, sampling and significance.

Preparation

Habitat: a beach, mud flat or other coastline of sand, mud or other fine loose material, preferably exposed at low tide.

Time required: 1 - 3 hours in the field plus 2 - 3 hours processing the samples in the laboratory or classroom.

Teacher: obtain references to local marine life. Select simple statistical tests for analysing the data.

Students: introduction to the kinds of marine life to be found, to types of organism distributions (regular, random, clumped) and to principles of sampling and statistical analysis.

Equipment:
- rope or string to mark transect
- quadrat sampling frame (square 25 cm x 25 cm or larger, or circle of plastic gardenhose up to 1 m diameter)
- rulers 20 or 30 cm
- plastic bags for samples
- buckets
- strong shovels or trowels
- sieves of different mesh sizes (e.g. 2 mm, 5 mm, 10 mm)
- pans or jars for sorting and observing organisms.

Strategy

This activity can be undertaken qualitatively (Question 1) and/or quantitatively (Question 2), depending on the level of the class and the time available.

Question: What is the zonation of the plants and animals on or in the sand or mud?

1. Lay out a transect across the intertidal from land to sea with 10 marks at regular intervals from the high tide line to the water's edge. Use a string or rope with evenly spaced marks, or use a tape measure and work across the area in segments.

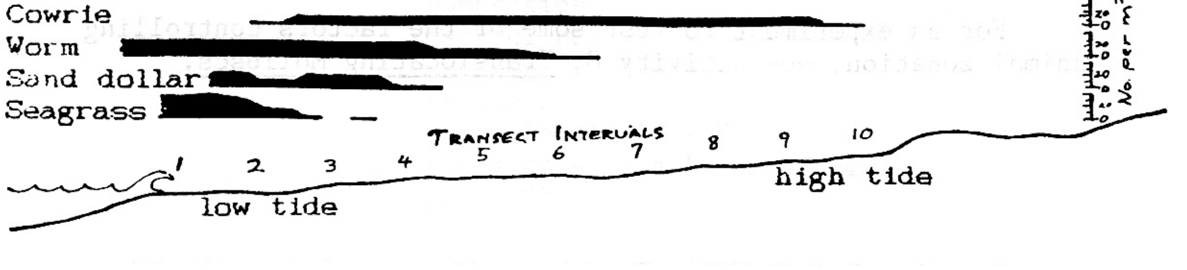
2. Starting at the seaward end at low tide, mark a quadrat sampling area at each mark using the square or circle frame, and note the kinds and numbers of all the organisms visible at the surface. Within the quadrat, or in a subsample of 25 by 25 cm if the quadrat is bigger, dig up the sand or mud to a depth of 10 to 20 cm together with any plants or animals in it. The sample can be washed through sieves immediately (see 3 below) or taken back to the school for washing and sorting. Keep each sample separate in a plastic bag, pan or bucket, and number the samples in order.

3. As you go along, or else later in the school laboratory, wash each sample through at least 2 sieves of different mesh sizes (say 10 mm and 2.5 mm). Note the kinds and relative abundance of organisms in each sample.

4. Prepare a chart showing where each organism occurs along the transect. In the example shown in Figure 31, the thickness of the line shows the abundance of each organism. Try to relate the distributions observed to known physical factors or gradients.

FIGURE 31

TRANSECT WITH ORGANISM ZONATION



Question 2: What is the distribution and abundance of organisms across the intertidal zone?

5. Lay a transect marked at 10 regular intervals from high water mark to the water's edge at as low a tide as possible, and take 5 samples at each mark at random distances from the line. Each sample should consist of the sediment and organisms in a 25 cm x 25 cm quadrat removed to a depth of 10 - 20 cm, and should be kept separate and numbered according to its location.

6. The samples should be sieved and sorted by kind of organism, and the numbers of each organism recorded for each sample. Prepare a table showing the distribution of organisms across the transect.

7. The significance of the differences observed between samples at the same mark, and between marks, can be tested statistically, either with manual methods or by processing the data on a microcomputer. Try to explain the differences in distribution and abundance between organisms.

Teaching hints

The size of the quadrats sampled will depend on the organisms present. For large organisms such as sand dollars, starfish and sea cucumbers, it may be necessary to mark off a 2 m by 2 m square, or even a 4 m radius circle defined by turning a rope around a stake. For small organisms (worms, small clams, etc.) the 25 cm quadrat may be sufficient, or an intermediate size (50 cm by 50 cm, 1 m square, or 1 m diameter circle) may be needed, depending on experience in the local environment.

On a coral reef lat, the underside of loose coral blocks can be a good sampling space. Measure the block and count all the attached organisms on the top and bottom. Standardise the counts per unit of area later.

A microcomputer equipped with data-base management (or spreadsheet) software can be used to construct tables and perform graphic analyses. Statistical tests like analysis of variance and regression analysis can also be done easily with standard commercial software.

Follow up

For an experiment to test some of the factors controlling animal zonation, see Activity 8, Translocating Molluscs.

3. THE MANGROVE COMMUNITY*

(A, B, C; a; I, II, IV)

Objectives

1. to observe and record the diversity and distribution of populations associated with gradients in physical factors.
2. to study interrelationships among organisms and between organisms and the physical environment.
3. to understand spatial relationships in the mangrove ecosystem.
4. to consider the effects of disturbing the mangrove ecosystem.
5. to understand the need for conservation of mangrove areas.

Curriculum links

Community, habitats, biological factors, physical factors.

Preparation

Habitat: a typical coastal mangrove area; if possible both undisturbed and disturbed mangrove areas can be studied and compared.

Time required: 2 - 3 hours of field work and a similar time for laboratory study of collections and data analysis.

Teacher: select appropriate sites for the surveys where the slope of the land can be measured in a straight line. Divide students into small groups (up to 4 - 5), with one group to measure the slope of the shore while the others make records at each station.

Students: a general introduction to mangrove forests would be useful. Special problems of working in mangroves, such as not walking in others tracks (because of the danger of sinking deeper in the mud) and keeping close to a supporting plant, should be understood.

Equipment:
- rope or string
- stakes
- plastic buckets (1 per group)
- plastic bags (5, 10, and 20 litres) and rubber bands/ties
- marking pen
- thermometer
- pH paper
- paper labels (3 x 5 cm)
- sieve or screen (1 mm gauge)
- jars (250 cc)

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*Based on an activity devised by Dr. Pisam Soydhurum.

scissors
forceps
small shovel
quadrat sampling frame (25 cm square or larger)
field notebooks and pencils
level or long clear plastic tube filled with water
measuring tape
ethyl alcohol (optional)
plant press (optional)

Strategy

Question: How are mangrove ecosystems and the organisms that live within them different from other ecosystems?

1. Lay a straight transect line with rope or string from the water's edge to dry land above high tide level. Place stakes at 10 metre intervals along the line to mark a station for each group of students. Number the stations.

2. To sample the animals at each station, throw the quadrat at random in the vicinity of the stake. Observe and record the kinds and numbers of animals found on the surface or underground within each quadrat. Put examples of each animal in one plastic bag, and a soil sample in another, and label them with the station number. Also record the kinds and numbers of animals or plants (especially mangrove roots) nearby. Throw the quadrat again and make another count and sample of speciments.

3. For the plants, mark off squares 10 x 10 metres along one side of the transect. Within the square, count and record the names and heights of all plants with a diameter of 4 cm or more. Collect samples of each plant, including leaves, flowers and fruits if possible. Record the animals found on each part of the plant.

4. Record the physical factors at the station:
   - characteristics of the soil (colour, odour, hard or soft)
   - light intensity (estimated qualitatively)
   - pH of the soil using pH paper
   - air temperature
   - soil temperature at 5 - 10 cm depth (allow 2 minutes)
   - slope or inclination (using tape measure and sight level or water level in transparent plastic tube).

5. After the field work or in the laboratory, wash the animals by running water over them in a sieve or screen. If a voucher collection is needed, preserve the animals in jars of alcohol; dry plant specimens in a plant press. Examine the plants and animals (with the help of a hand lense or microscope, if available), and identify them if possible.

6. Prepare a table listing for each station the name, density (number per square metre), height (for plants) and observable characteristics of each animal and plant on the surface or underground.

7. Make a table for the physical factors, showing for each station the soil characteristics (colour, odour, soft or hard), soil temperature, air temperature, pH and light intensity.

8. Draw a diagram showing the slope of the area studied in cross-section, and enter the information from the table of plants and animals to show their distributions along the transect in order to demonstrate the spatial distributions in the ecosystem. (Figure 32)

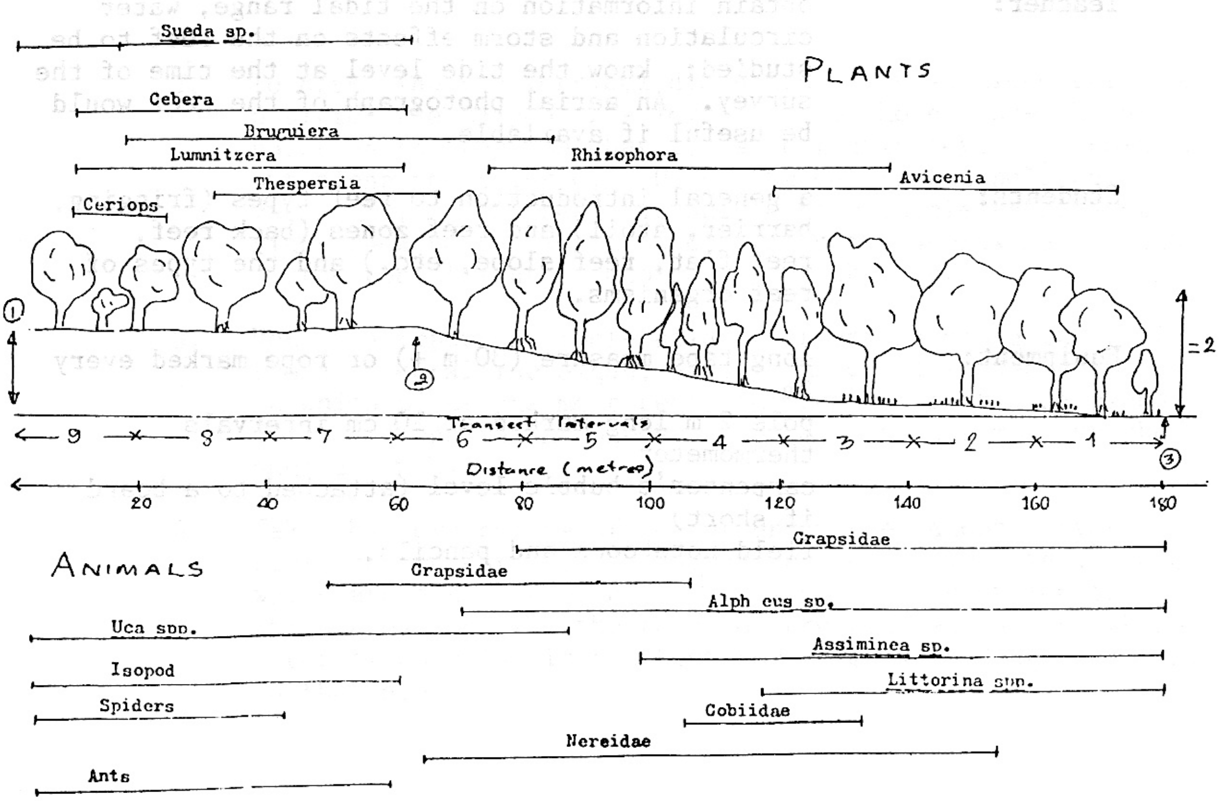
9. If a disturbed mangrove site is available, do the same survey in the disturbed site and compare the two transects.

10. Discuss with the class what may happen with different kinds of mangrove disturbance (logging, filling, polluting, changing water circulation).

11. Review with the class the values of mangroves and the importance of their conservation.

FIGURE 32

PLANT AND ANIMAL DISTRIBUTIONS IN A MANGROVE ECOSYSTEM



Follow up

The total picture of the mangrove ecosystem formulated from the data in the field activity could be compared with data from other classes or from other areas. Any similarities or differences should be discussed.

4. CORAL REEF STRUCTURE

(A, B; I, II)

Objectives

1. to observe the structure of the coral reef.
2. to relate the reef structure to some environmental factors.
3. to study the role of reef-building organisms.

Curriculum links

Community structure, organism-environment relations (biological and physical factors), skeletal accumulation.

Preparation

Habitat·	an accessible coral reef where pupils can observe as many different reef zones as possible in safety; to be surveyed at a very low tide.
Time required:	2 hours in the field.
Teacher:	obtain information on the tidal range, water circulation and storm effects on the reef to be studied; know the tide level at the time of the survey. An aerial photograph of the reef would be useful if available.
Students:	a general introduction to reef types (fringing, barrier, atoll) and reef zones (back reef, reef flat, reef slope, etc.) and the types of reef organisms.
Equipment:	long tape measure (30 m +) or rope marked every 2m pole 2 m long marked at 10 cm intervals thermometer carpenter's bubble level (attached to a board if short) field notebooks and pencils.

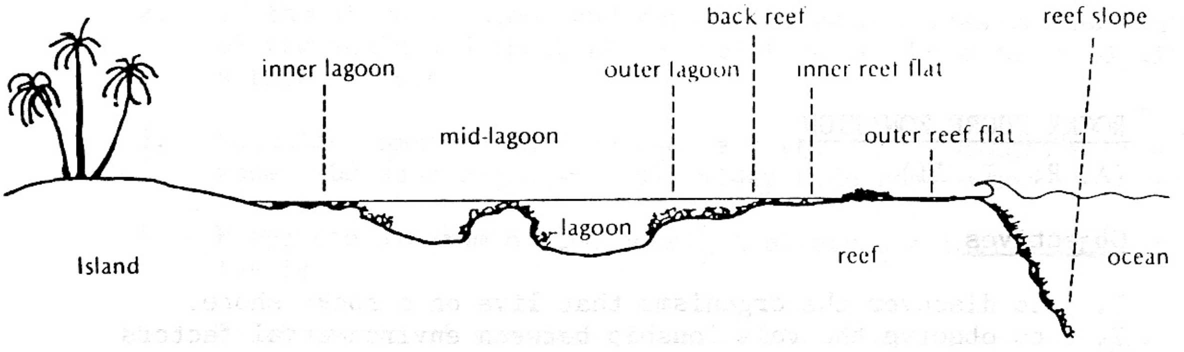
Strategy

Question: What is the structure or form of the coral reef?

1. The best way to visualise the structure of a coral reef is to draw a vertical profile (Figure 33). Select a typical area of coral reef, and establish a starting point on the shore. Lay out the tape measure or marked rope seaward from the starting point, and follow this transect line out recording the type of bottom and organisms and their distance from the starting point. It may be necessary to move the tape or rope several times to cross the whole reef.

FIGURE 33

CORAL REEF PROFILE WITH ZONES



2. To determine the vertical dimension (depths) along the profile, position the carpenter's level on the shore above the highest point on the reef so that it is perfectly level and pointed out along the transect. Using the calibrated pole held vertically on the reef, it should be possible by line of sight back to the level to determine the approximate depth of the reef features recorded along the profile.

3. Note the important reef-building organisms along the profile (corals, coralline algae, etc.). Have the students observe in which zones the most reef construction is taking place. Try to recognise the pieces of organisms that make up the sand and rubble.

4. Take the temperature of the water in the lagoon, beyond the reef, and in any pools of water trapped at low tide. Note which areas are exposed to air, sun and rain at low tide, which areas receive the force of breaking waves, and where water circulation is the best for animals feeding on plankton in the water.

5. Have the students imagine what happens on the reef during a major storm. Is there evidence of storm damage, such as large rocks, rubble banks or other accumulations on or behind the reef?

6. From the measurements and observations made, the class should prepare a profile of the coral reef, labelling the zones observed and relating them to physical factors. As many pertinent details as possible can be added to the profile.

7. If an aerial photograph of the reef is available, relate the profile of the reef to its overall shape and note the distribution of reef zones.

Teaching hints

The preparation of the profile can be an individual or group exercise. If the class has access to different types of reef, profiles of these different types can be made and compared.

5. ROCKY SHORE ZONATION

(A, B; I, II)

Objectives

1. to discover the organisms that live on a rocky shore.
2. to observe the relationship between environmental factors or gradients and organism distributions.
3. to look for examples of competition for space.

Curriculum links

Identification of organisms, distributions, gradients of environmental factors, zonation, competition.

Preparation

Habitat:	a rockyshore, seawall, wharf piling or other hard substrate across the intertidal zone and accessible at low tide.
Time required:	up to 2 - 3 hours, depending on the width of the intertidal.
Teacher:	obtain references to local marine life and tidal cycles.

Students: introduction to the organisms of rocky shores; explanation of tides and their effects on exposure in the intertidal.

Equipment: tape measure
field notebooks and pencils

Strategy

Question: What is the distribution of marine life in the intertidal zone?

1. Many organisms try to crowd onto a rocky shore where they can attach and receive light, water and nutrients. They usually occur in horizontal bands or zones in the intertidal zone depending on how often and how long they are exposed to air. The class should follow a low tide out and in, observing and making notes on the zones of organisms observed.

2. If the shore is steep and not too irregular, measure the width of the zones and their height relative to the tide level with a tape measure.

3. Note the common organisms that are the best indicators of each zone. Do some organisms have wider tide ranges than others?

4. Prepare a diagram of the zonation observed relative to tide levels.

5. On the basis of the observed zonation of organisms and the information on tidal cycles, the class should determine the length and frequency of exposure to the air for each zone identified.

Follow up

The activities on functional morphologies can be used following this activity to explain some of the adaptations that allow organisms to survive the conditions in each zone.

6. ZONATION ON MANGROVE TRUNKS

(A, B, C; I, II)

Objectives

1. to discover the organisms that live on mangrove trunks and prop roots.
2. to observe the relationship between environmental factors or gradients and organism distributions.
3. to appreciate the importance of a place to attach in an environment where attachment is difficult.
4. to observe how organisms can create an environment for other organisms.

Curriculum links

Identification of organisms, distributions, gradients of environmental factors, zonation, interrelationships.

Preparation

Habitat: mangroves with accessible trunks or prop roots well colonised by benthic organisms.

Time required: 1 - 2 hours of field observations.

Teacher: select a good low tide when trunks will be accessible; obtain references to local marine life.

Students: an introduction to mangrove ecology; an explanation of tides and their effects on exposure in the intertidal.

Equipment: Knife or scraper for collecting
bucket, plastic bags or small jars
metre stick or 2m tape measure
field noteboosk and pencils
hand lens or microscope

Strategy

Question: What organisms live on mangrove trunks and how are they distributed relative to the tide level?

1. The soft, muddy bottoms of mangrove coastlines are not good places for many plants and animals to attach. Mangrove trunks and prop roots are often the only place where algae can stay in the light and animals keep out of the smothering mud. Such places are thus often crowded with life. However, the rise and fall of the tides covers and uncovers different levels of the trunks for different lengths of time, producing a vertical zonation of organisms.

2. Measure the width of each zone or band of organism distribution relative to the tide level.

3. Collect samples of each organism or association for identification on the shore or in the laboratory. Examine the algae and small animals with a hand lens or microscope. Determine the names of as many organisms as possible.

4. Combine information on the levels of each zone and on the tidal cycle to calculate the frequency and duration of exposure for each zone.

5. Prepare a diagram showing the zonation of common organisms on a mangrove trunk relative to the tide levels.

6. Try to explain the distribution of organisms in terms of their resistance to exposure to air, their vulnerability to predation by animals in the water, or other factors.

7. If there are few or no organisms at certain levels, try to explain why (changing heights of the sediment, grazing, pollution, storm scour, sedimentation, turbidity, etc.).

8. If time permits, experiment with the effects of changes in level on organism distributions. Select three similar prop roots or small trunks with the same zonation patterns. Cut off two of them above and below the main intertidal zones, and reattach them to one side of the third (the control), displacing one 20 - 30 cm (or more if the zones are wide) above its normal level, and the other the same distance below its normal level. The roots can be wired or nailed into place.

   Return to examine the changes in organism distributions and zones on the two displaced roots relative to the control, after 1 day, 3 days, 1 week, etc. until the normal zonation is re-established. Measure and note the changes observed on each visit. How long does it take each organism to readjust to its normal zone? Do some move while others disappear and grow back again? Do some organisms persist outside of their usual zones? How do you explain these observations?

7. AREA COVERAGE ON A CORAL REEF

(A, C; c, d: II)

Objectives

1. to observe the area of a coral reef covered by different types of plants and animals.
2. to learn the importance of semi-quantitative observations.
3. to estimate the relative importance of different groups in productivity and reef construction.

Curriculum links

Community structure, productivity, competition for space, environmental impact assessment.

Preparation

Habitat: choose a shallow coral reef area, preferably not steeply sloping, with good coral cover for the area, such as a reef flat or shallow back-reef.

Time required: 3 hours.

Teacher: the categories of bottom and organisms to be quantified may need to be adapted or simplified for local conditions.

Students: the categories to be measured and the method for estimating percent cover should be studied carefully before the field activity.

Equipment:
- steel stakes or rods 1 m long
- heavy hammer
- ropes or cords with loops at each end, 4 m long
- or tape measures 30 - 50 m long
- field notebooks or underwater slates and pencils
- face masks or goggles (snorkles and fins desirable)

Strategy

Question: How important are the different plant and animal groups on a coral reef?

1. Divide the class into groups of 2 - 5 students. One or more students shoudl make observation while another records the results. Measures by different students can be recorded separately and cross-checked for consistency. Select one survey site for each group, spaced some distance apart. Two alternate methods can be used to estimate percent cover, as follows:

2a. **Estimation within a circle.** Drive a steel stake into the centre of each survey site. Put one loop of the 4 m rope over the stake. The circle described by the rope as it pivots around the stake defines a survey area of 50 square metres. Observing the bottom as the rope passes over it helps to make observation of the area more systematic. The estimation of the percent cover, or area of the circle occupied by either a class of sediment or a group of organisms, requires a certain educated judgement. Six classes coded 0 to 5 can be used:

0 = none (0%)
1 = a little (1 - 5%)
2 = some (6 - 30%)
3 = nearly half (31 - 50%)
4 = more than half (51 - 75%)
5 = almost all (76 - 100%)

It is difficult to make accurate judgements for the area covered by many small scattered things, a few big things, and things that are unevenly distributed. Figure 34, page 87, illustrates some different amounts and types of area coverage.

2b. **Line intercept method.** A tape measure 30 to 50 metres long is laid out parallel to the reef or shoreline. The group goes along the line recording the number of centimetres covered by each type of bottom, and then a second time recording the types of organisms. The total number of centimetres for each group, divided by the length of the line, gives the percent coverage for that group. If possible there should be enough tape measures for each group to have their own line, or several lines can be done sequentially and compared.

3. In looking at the bottom, there will be sand or bare rock in some places, and corals, plants or other things that are attached or fixed in place on the bottom. Two important measures of area coverage will be made: bottom type and major biological groups.

4. The first measure is what the bottom is made of. Much of a reef is solid, hard rock all cemented together, but in some places, and particularly on the lagoon floor, there is loose rock or sand on top of this. Measure what is seen at the surface, not what may be underneath if you dig down. The loose material or sediment is subdivided into 4 types based on the size of the individual grains, giving 5 categories of bottom type:

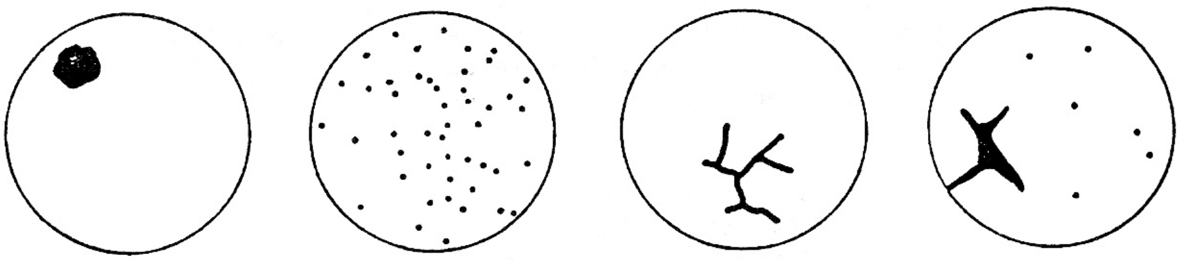
mud: very fine and soft, individual particles not distinguishable;

sand: small bits of rock or shell with the separate grains visible, but smaller than rubble;

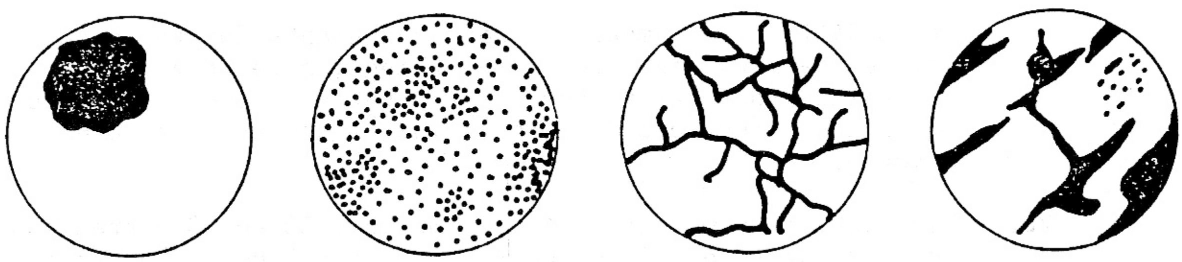
FIGURE 34

CLASSES OF PERCENT COVER

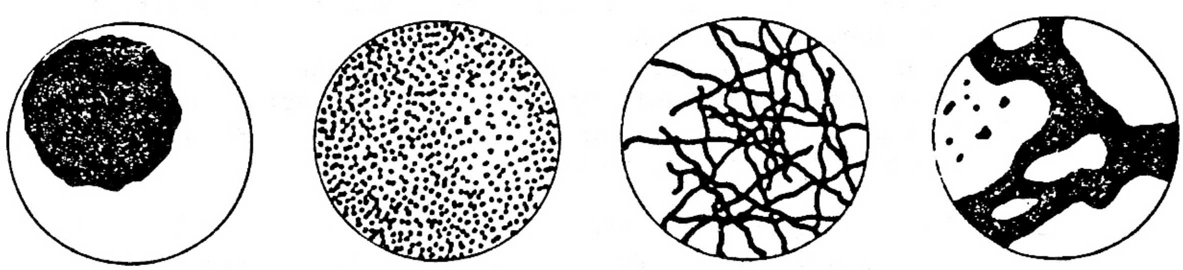
1 = 1 – 5% : a little



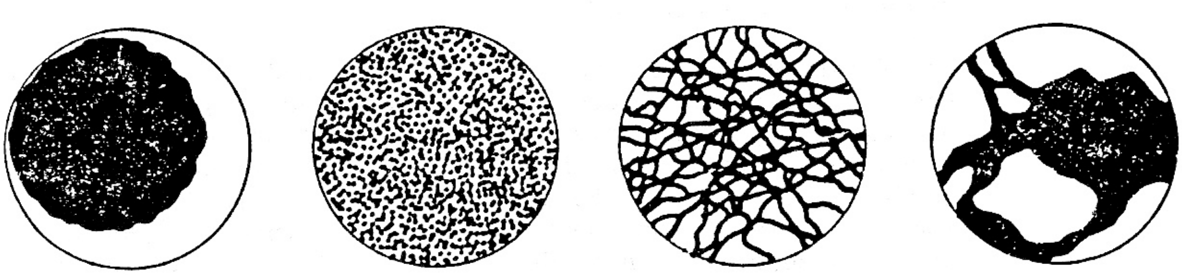
2 = 6 – 30% : some



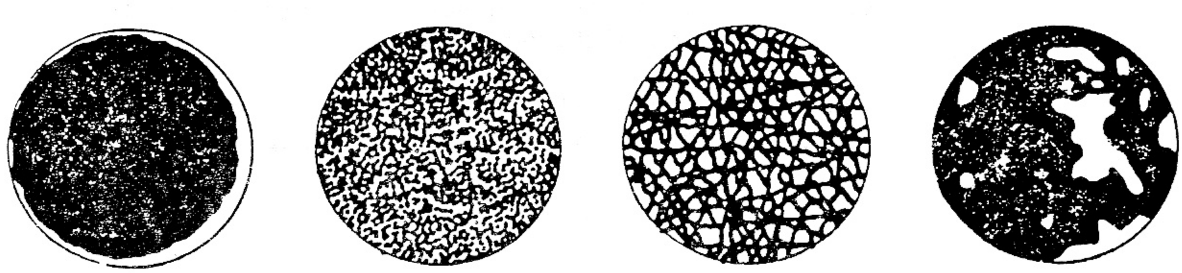
3 = 31 – 50% : nearly half



4 = 51 – 75% : more than half



5 = 76 – 100% : almost all



rubble: pieces of broken coral or rock ranging from the size of a finger joint up to the size of a human head;

blocks: coral rocks larger than a human head but still not attached or cemented to the bottom.

rock: the solid rock of the reef.

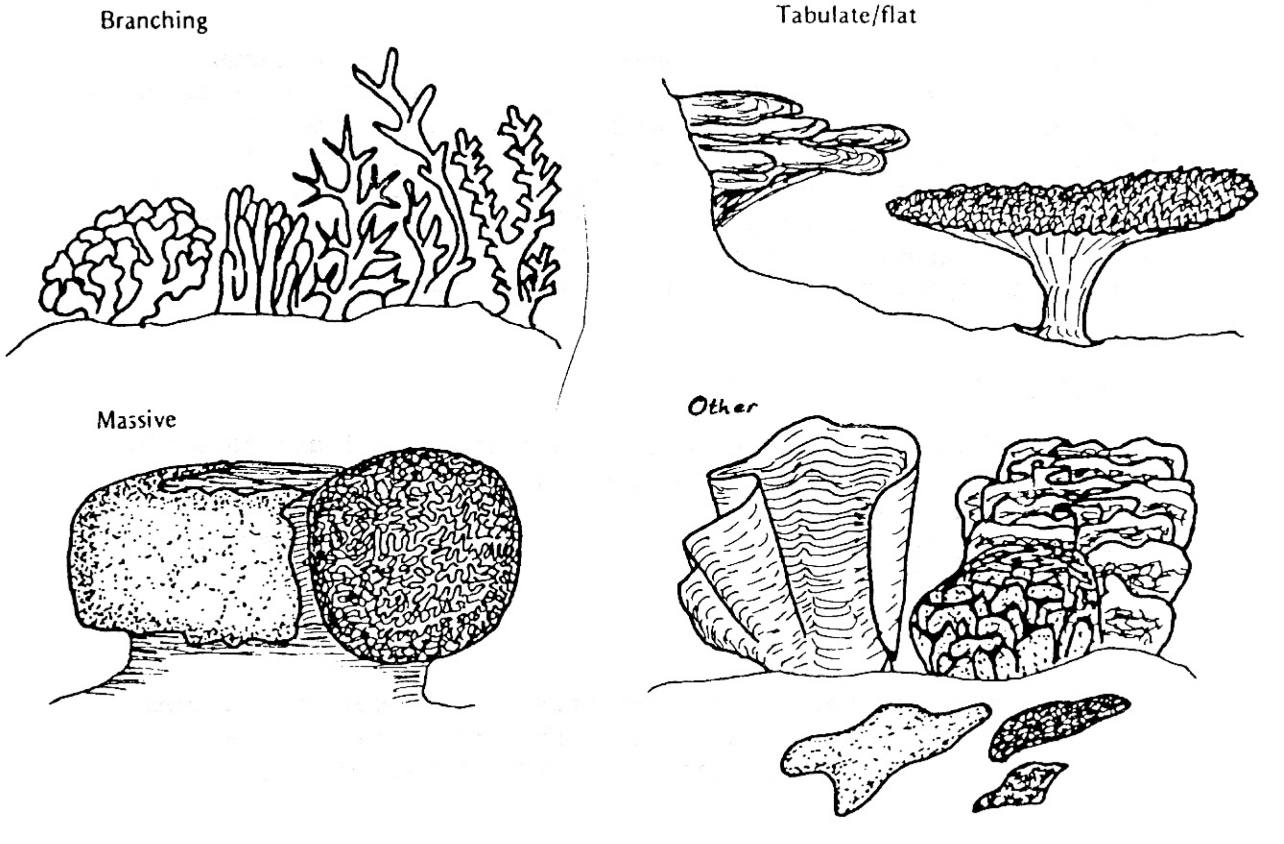
Each group should estimate and record the percent cover of each of these types of bottom in the circle. The sum of all the types should equal 100%.

5. The second measure is the amount of bottom covered by major biological groups attached to it. The following categories are distinguished:

live hard corals: these corals feel hard because they have a stony skeleton, and are covered by the tiny animals or polyps that live in holes in the skeleton. The holes may be like tiny craters, pin-holes or valleys and are often rough or sharp around the edge. The corals generally look "clean" and are coloured by the animals tissues. They may be of many shapes: big like boulders, small and branched like bushes, or just a crust on the rock. Live hard corals can be subdivided into 4 major forms: **branching, massive, tabulate/flat,** and **other**. (Figure 35)

FIGURE 35

FORMS OF LIVE HARD CORALS



soft corals and sponges: these may have shapes like corals but they are soft to the touch, sometimes slimy or rough and flexible. Some are very brightly coloured. If there are waves, they may move back and forth.

dead standing coral: when hard coral dies, only the white skeleton is left. Fuzzy plants may then grow on it, giving it a dirty brown, blackish, pinkish or reddish colour, but the outline of the coral skeleton is still very clear and it is still standing in the place where it grew. This is dead standing coral, and it has not died too long ago, showing that something has killed corals on the reef. Eventually the skeleton is broken up, eroded or overgrown by other things and becomes part of the reef rock and rubble, and is no longer dead standing coral. if it is not clear that something is dead standing coral, do not count it.

crustose corallines: these are plants (algae) that have a stony skeleton-like corals and grow as a crust on dead parts of the reef, cementing it together. They range from dark pink or purple to whitish-yellow in colour (grey in deeper water) and have a smooth or lumpy surface like spilled paint.

marine plants: these include the sea grasses and seaweeds and can range from a cushion-like turf to big floppy plants. They may begreen, red or brown in colour. Only plants or clumps of plants large enough to grab hold of should be counted; fine algal films and fuzzes are too difficult to measure.

Each student group should estimate the percent coverage of each of these categories. The difference between the total of these percentages and 100% is the part of the reef without any obvious organisms.

7. The class should discuss the results of the survey in terms of the importance of each type of bottom to the construction or erosion of the reef. The coverage of organisms should be related to reef processes such as reef construction, food production, and the creation of shelter for fish and other important animals. A low coverage of live hard corals and coralline algae shows a reef that is no longer growing. A significant amount of dead standing coral is a sign that the reef has been damaged recently.

Teaching hints

Care should be taken that the class does the least possible damage to the reef. Walking on a reef, standing on or grabbing corals can cause significant breakage of fragile species. Swimming over the reef is less damaging, if that is possible.

Follow up

This activity can be repeated over time to observe changes in the reef, or used to compare different reef areas. See activities 32 (Monitoring) and 33 (Environmental Impact).

Reference

This is a simplified version of techniques described in the Coral Reef Monitoring Handbook, United Nations Environment Programme, Reference Methods for Marine Pollution Studies No. 25 (1984), available from UNEP OCA/PAC, P.O. Box 30552, Nairobi, Kenya. The whole handbook is suitable for use at secondary school level.

8. TRANSLOCATING MOLLUSCS

(A, C, D; a; III)

Objectives

1. to experiment with factors controlling animal distributions.
2. to learn the value of experimental approaches to ecological research.

Curriculum links

Animal distributions, limiting factors, population dynamics.

Preparation

Habitat: any intertidal area (rocky shore, sand flat, mangrove trunk) where a species of mobile mollusc, such as a snail or periwinkle, is common in a narrow band or zone.

Time required: two visits of 2 hours each one to two weeks apart.

Teacher: select an area where a common local intertidal mollusc is suitable for the experiment. If possible, obtain some information on the ecology of the species concerned (food, predators, resistance to exposure, preferred ecological conditions) to use in explaining the experimental results to the class. Verify that the marking technique used to identify the snails is sufficiently persistent that loss of marks does not affect the result.

Students: should understand the idea of an experiment and a control.

Equipment: tools for collecting molluscs (shovel, sieve or knife may be necessary depending on the species and habitat).
Fast-drying waterproof paint, nail polish, or permanent marking pens (3 colours)
3 stakes or other means of marking the experimental site.

9. CORAL REEF SURFACE AREA

(A, C; I, II)

Objectives

1. to observe the three-dimensional structure of a coral reef.
2. to relate the structure to environmental factors.
3. to learn the functional importance of surface area.
4. to understand the choices involved in different life-form strategies.

Curriculum links

Functional morphology, community structure, photosynthesis.

Preparation

Habitat: choose a healthy coral reef with as much living coral and vertical relief as possible.

Time required: 1 - 5 hours on the reef and 1 - 5 hours in the classroom.

Teacher: if the students do not have a background in solid geometry, they will need to be provided with the formulae for calculating the surface of such forms as a circle, triangle, rectangle, sphere, cone, and cylinder.

Students: some ability in mathematical operations such as calculating the surface of a sphere or cylinder.

Equipment: measuring tapes, metre sticks, rulers
40 m of rope or cord marked at 10 m intervals
4 stakes or rods
field notebooks and pencils
calculators

Strategy

1. The surface of an organism has great functional significance. It determines the amount of light a plant absorbs, the uptake or loss of gases, water, nutrients, and heat, and then drag that must be resisted in a wind or current, among other things. An ecosystem such as a coral reef also creates surface as it becomes richer. In this activity students will observe how much surface area a coral reef can make, as a basis for understanding its ecological signfiicance.

2. Select a healthy coral reef area with many corals. Mark off a quadrat (square) 10 m on a side with the rope and stakes. Horizontally this quadrat covers 100 square metres.

3. Measure the size of the gross relief of the bottom, such as the size of ridges or depressions, and prepare a sketch map of the quadrat, noting the dimensions measured.

4. List or note on the map the numbers and sizes of the major corals. For each type of coral, measure its dimensions (height, diameter, circumference, number of branches, diameter of branches, distance between points of branching, etc.). Other fixed animals should be counted and their size recorded.

5. Make similar counts and measurements for significant plants in the quadrat. Measure the approximate area covered by plants, the height of the plants, the number of blades or fronds or their average density per unit area, and the size or diameter of the blades.

6. These measurements will be used in the classroom to reconstruct the quadrat with simple geometric forms that approximate to the forms observed on the reef, but for which it is easy to calculate the surface area. For instance, ridges on the reef may resemble a corrugated surface made up of half cylinders; a massive coral may be approximated by a hemisphere of the same dimensions; a branching coral by a set of cylinders branching in the same way (Figure 36 , page 97). The pupils can use their imaginations to recreate theorganisms observed using geometric forms, and then calculate the surface area of the forms. First calculate the surface of each type of coral or other organism at the average size observed, then multiply by the number observed to get the contribution of that organism to the total surface of the quadrat. The total area of the reef surface plus the corals and other organisms gives a surface area index, which can be expressed in square metres of surface for each horizontal square metre of bottom. A reef with many corals might have a surface area index of 15, so the 100 square metre quadrat may have 1500 square metres of functional surface. A moderately well developed reef might have half of that, and a poor reef an index of less than 3.

7. Discuss the importance of the surface observed for such reef processes as absorbing light for photosynthesis, feeding on plankton, exchanging nutrients and waste products with the water, resisting storm waves, etc.

Teaching hints

This activity will be particularly meaningful if it follows the study of photosynthesis or other biological processes for which surface is important.

Strategy

Question: Can an organism survive outside its usual zone of distribution?

1. Select a site where the mollusc is common in one zone but not in adjacent areas above or below the zone. Collect 60 to 150 of them, divide them into 3 groups of 20 to 50, make sure the shells are clean and dry, and mark those of each group with paint of a different colour. The sample size chosen will depend on the numbers of animals available and the time needed to apply the marks.

2. Place three markers (paint can be used on hard surfaces, stakes in soft substrates), at the place where the molluscs were collected, and in zones seaward and landward of that zone (but not too far away) where they do not occur.

3. Release one group of 20 - 50 (the control) in the immediate vicinity of the marker where they were collected, and the other groups around the other markers. Position them so that they can reattach or rebury themselves if possible.

4. Return a few days or a week later, examine the surface or sieve the sand around each marker, and count the number of organisms in each group that can still be found.

5. The numbers of surviving translocated organisms should be compared to the number of control organisms (returned to their original location). What percentage of marked organisms can still be found? How far have the marked organisms moved? Have any transplanted organisms returned to their normal zone?

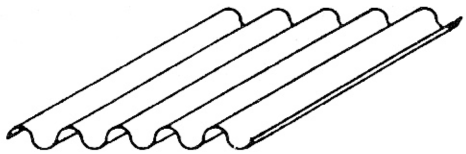
6. Discuss with the class the environmental factors that could explain the results observed, and possible interpretations in terms of movement, mortality rate and population size.

FIGURE 36

GEOMETRIC FORMS APPROXIMATING REEF SURFACES

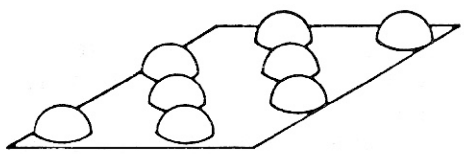
THEORETICAL REEF SURFACE

SCALE I — GROSS REEF MORPHOLOGY

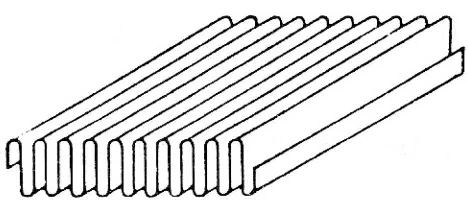


RIDGES 1 M HIGH AND
1 M APART

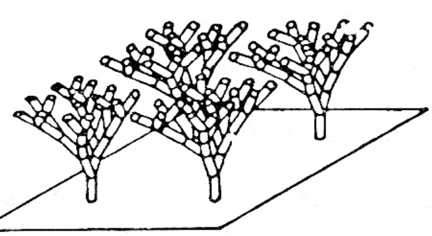
SCALE II — INDIVIDUAL CORALS



25% MASSIVE AND CRUSTOSE
.25 M DIAMETER
HEMISPHERES, 8 PER M^2



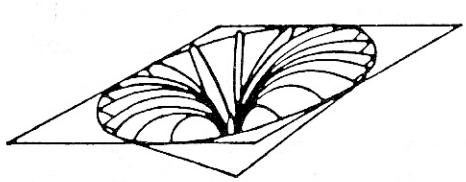
25% PLATE-LIKE
RIDGES .25 M HIGH, .05 M
WIDE, AND .05 M APART



25% BRANCHED
CYLINDERS .5 M HIGH, .05 M
DIAMETER, BRANCHED EACH
.1 M, SPACED .1 M AT TIPS

25% NO RELIEF AT SCALE II

SCALE III — CORAL OR ROCK SURFACE



60% CORAL SURFACE
POLYP CUP CONICAL 10 MM
DIAMETER, 5 MM DEEP, WITH
20 SEMICIRCULAR PLATES
2.5 MM HIGH

40% NO RELIEF AT SCALE III

Follow up

It is possible to use this activity to compare different reef zones, reefs exposed to different environmental conditions (such as windward and leeward reefs, or exposed and sheltered reefs). or undisturbed reefs and those that have been damaged by human activities.

Reference

A more complete explanation of surface area estimations on coral reefs can be found in: Dahl, A. L. Surface area in ecological analysis: quantification of benthic coral-reef algae, Marine Biology, Volume 23, 1973, pages 239 - 249.

10. REPLANTING MANGROVES

(C; a, b: III, IV)

Objectives

1. to learn the techniques for replanting mangroves.
2. to see how to experiment with the re-establishment of more productive or natural communities.
3. to appreciate the value of restoring exploited or damaged ecosystems.

Curriculum links

Mangroves, reforestation, restoration of degraded ecosystems, environmental management.

Preparation

Habitat: a former mangrove area where the trees have been damaged or removed but suitable shallow muddy habitat exists.

Time required: 2 - 3 hours of field work plus occasional subsequent visits to observe the results.

Teacher: select a suitable coastal area in need of reforestation with mangroves. Be certain that the conditions that caused the loss of mangroves have been corrected. Identify also an existing mangrove area from which propagules (seedlings) can be collected at a stage suitable for planting. Choose a low tide period when the site to be planted is accessible.

Students: an explanation of mangrove reproduction and dispersal should be given before the field activity.

Equipment: knife for collecting propagules
buckets or plastic bags
small shovels or trowels.

Strategy

Question: How can a mangrove forest be re-created where it has been damaged or lost?

1. Construction, pollution or exploitation can damage or destroy a natural ecosystem that may be valuable for man. We cannot go on destroying nature for ever, so we must learn how to help natural systems grow back orrecover after the damaging influences have been removed. Not much is known about restoring damaged marine ecosystems, but mangrove forests have been replanted in many places. Since mangroves are important for coastal protection, and for the food and shelter they provide many commercially important species, they should be restored whenever possible. This activity will show how to do it.

2. Study your local mangrove community to see the conditions under which each species lives (Activity 3, page 79). Observe the propagules being produced on the trees, and the young seedlings that have fallen, found a suitable site, and started to grow. Measure how far apart the adult trees are spaced.

3. Study the damaged area to be replanted in the same way to identify the parts of the area with conditions appropriate for each species.

4. Collect enough mature propagules to cover the area to be planted at slightly more than the same density as in the natural area (to allow for some mortality).

5. Mark out the spacing in the area to be planted, and plant the propagules at about the same depth as that observed in nature. Be certain that each species is planted in its preferred conditions. Count the number of propagules planted and measure their heights at planting.

6. Return at intervals of 2 to 4 weeks to see how the new seedlings are doing. Count and measure the heights of the survivors.

7. Make graphs of the results, including the number of seedlings surviving over time, and the growth (average height) of the seedlings over time.

Teaching hints

Mangroves reproduce by producing propagules, which are basically seeds that sprout and start growing on the tree, so that when they drop they can root quickly in the mud and not be carried away to sea. Learn to recognise when the propagules are ready for planting.

If many of the seedlings are lost soon after planting, check that conditions in the area are suitable. A storm that washes away new plants is a hazard that perhaps cannot be totally avoided, but waves and erosion are also a danger.

Starting the seedlings in a protected "nursery" area and then transplanting them once the roots have started to develop may improve their holding power in an exposed site. If the seedlings die soon after planting, the problem may be pollution or unsuitable salinity (construction frequently changes drainage patterns, killing mangroves). If the planting activity fails, it will at least illustrate how difficult it is to recreate natural areas.

Follow up

A successful mangrove planting project can be a good demonstration for more widespread environmental restoration by local communities or governments.

If this becomes an annual class project, the class can also visit sites planted in previous years and compare them with undisturbed natural areas to observe the regeneration of the ecosystem.

11. TROPHIC LEVELS

(B; a; I)

Objectives

1. to learn the different tophic levels.
2. to observe which local organisms belong to each trophic level.
3. to understand the prey-predator relations between organisms.

Curriculum links

Trophic levels, food chains, biomass and energy flow, predator-prey relations, feeding adaptations.

Preparation

Habitat: any intertidal habitat, preferably with attached plants because primary production based on filter-feeding on plankton is more difficult for pupils to visualise.

Time required: 2 hours in the field plus 1 hour class discussion.

Teacher: learn the feeding habits of local organisms. If time is limited, prepare a list of organisms, possibly illustrated with simple drawings, which pupils can use to note their observations on trophic levels.

Students: should understand the different trophic levels (primary producers, herbivores, carnivores, omnivores, detritus feeders, decomposers) and the concept of food chains or food webs.

Equipment:
field notebooks and pencils
knife or prying tool
magnifying glass or hand lens
face masks for underwater observations
dissecting tools (small knife and forceps)
dissecting microscope (optional)

Strategy

Question: What are the plants and animals that make up the coastal food web?

1. Visit the sea shore at a low tide when as many organisms as possible are accessible.

2. Each organism should be observed closely, and on the basis of its appearance and morphology (plant or animal, fixed or motile, type of mouth parts, location and observed behaviour, etc.) the students should assign it to an appropriate trophic group:

   - primary producers: plants and animals with symbiotic algae;
   - herbivores;
   - carnivores (can have several levels)
   - detritus feeders and decomposers

   Note that some organisms may belong to more than one groups.

3. Probable prey organisms or sources of food should be identified.

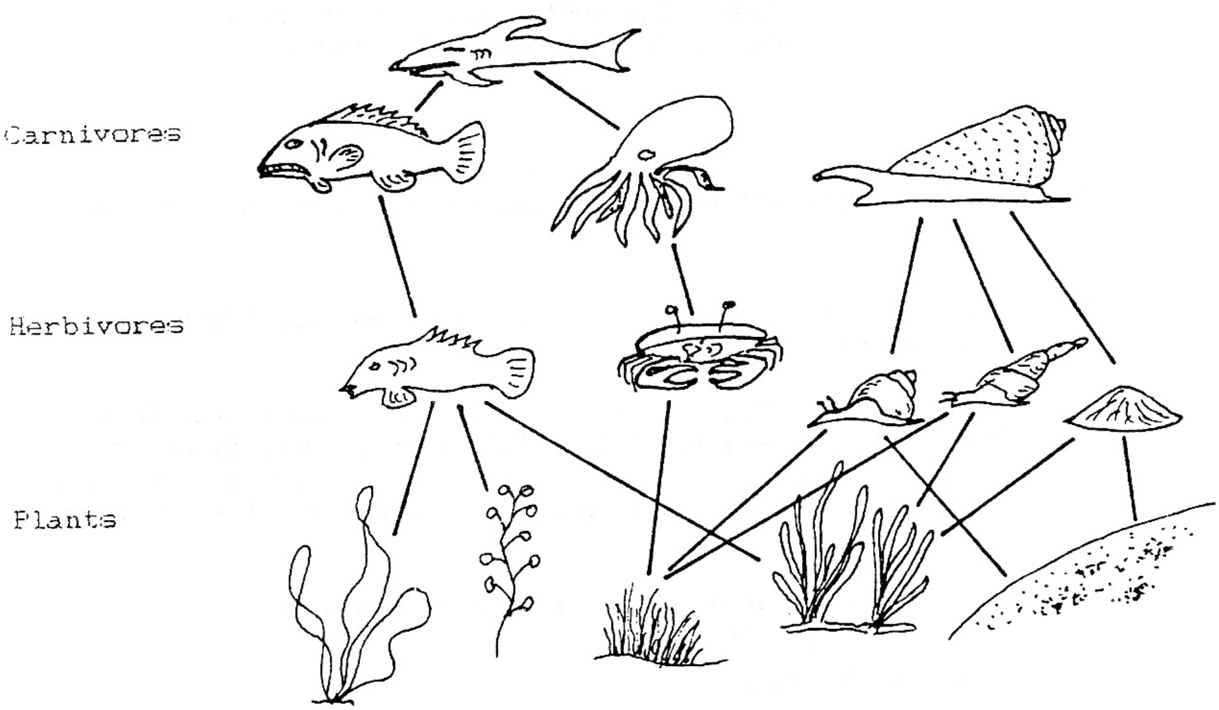
4. If the food of an organism is not apparent, the organism can be collected for examination in the classroom or laboratory using a hand lens or dissecting microscope. Mouth parts can be dissected, and stomach contents examined under a microscope (if available).

5. Any information the students may have on higher levels of the food chain, such as the fish they eat at home, and what those fish eat, can be added to their field observations.

6. On the basis of these observations, the class should construct a food chain or food web for the coastal community they have studied. (Figure 37)

FIGURE 37

EXAMPLE OF A MARINE FOOD WEB



Follow up

This activity can be followed by Activity 24 (page 126) on feeding adapatations.

12. SEAGRASS TURNOVER

(B, C; d; III)

Objectives

1. to observe primary production and loss (turnover) in seagrasses;
2. to use a field experiment to determine change over time.
3. to appreciate the importance of seagrasses in food production in coastal waters.

Curriculum links

Primary productivity, herbivores, detritus, food chains.

Preparation

Habitat:	shallow bottom with seagrass bed accessible to students.
Time required:	two visits of 1 - 2 hours each one week apart.
Teacher:	no special preparation required.
Students:	introduction to the ecological importance of seagrasses.
Equipment:	stiff wire stakes waterproof tags marked with group numbers or student names (can be cut from plastic food containers) 30 cm ruler or measured strings with loop at one end cheap staplers, paper punches or scissors (will rust) field notebooks or waterproof slates and pencils

Strategy

Question: How much food is produced by seagrasses?

1. Each student or small student group should choose a small area of dense seagrass and mark it with a stiff wire stake and attached plastic label driven into the sand. These markers should be left in place for the return visit.

2. The area to be measured is determined by turning the ruler or measured string around the marker. A 30 cm radius is good, but this can be increased or decreased depending on the density of seagrass blades (leaves). There should be 20 to 50 blades in the area measured.

3. Every seagrass blade touched at its base by the string should be counted.

4. All the blades that are counted should be marked in the middle, either with a staple (from an ordinary paper stapler), by punching a small hole in the blade with a paper punch, or by cutting a small notch in one edge with scissors. This will allow the old leaves to be identified on the return visit.

5. The class should return about 1 week later and relocate their markers.

6. Using the same string, they should count both the number of marked seagrass blades remaining and the number of new leaves without marks.

7. From these data, the density of seagrass leaves should be calculated. The number of new leaves is a measure of primary productivity (new food being made). The number of leaves originally marked minus the number of marked leaves remaining after 1 week gives the number of leaves grazed off by herbivores or lost as detritus to supply the detritus food chain.

8. Discuss the significance of the class results for the ecology of the area.

13. MANGROVE LEAF PRODUCTION

(B, A; II)

Objectives

1. to measure the production of organic matter by mangroves.
2. to use a simple quantitative field technqiue.
3. to appreciate the importance of mangrove productivity to coastal ecosystems.

Curriculum links

Mangroves, primary productivity, food chains, detritus, critical habitats.

Preparation

Habitat: any mangrove forest or coastal fringe.

Time required: 1 - 2 hours of initial field work followed by daily visits and 1 hour of laboratory work for the three or four succeeding days.

Teacher: select an accessible mangrove area with a reasonably high canopy density and leaf fall rate, in an area where the leaf traps will not be disturbed.

Students: an understanding of the mangrove detrital food chain.

Equipment: pieces of cloth 1 m x 1 m (such as unbleached muslin)
strong string or cord
laboratory balance or scales (accurate to 0.1 g)
drying oven (if available)

Strategy

Question: How much food is made by mangroves?

1. Mangrove leaves are the beginning of an important detrital food chain in the coastal environment, often extending to lagoons or coastal waters beyond the mangrove forest itself. Important commercial species may depend for part of their life cycle on food energy provided by decaying bits of mangrove leaf and the bacteria that feed on them.

2. To measure the amount of leaf material produced by mangroves, make leaf traps under the mangrove trees. Take 1 square metre of cloth, tie a strong string to each corner, and stretch it horizontally under the mangrove canopy by tying the strings to appropriate trunks or branches. Be sure the traps are high enough that they will not get wet at high tide. Place a rock or other weight in the centre of the trap to give it a funnel shape and to keep the wind from shaking the leaves out of it. Note the location of each trap and the species of mangroves over it. If possible make several leaf traps under each important mangrove species.

3. Return each day for 2 or 3 days (or at whatever interval is practical), remove all the leaves form each trap, put them in separate plastic bags labelled as to the trap they come from, and return them to the laboratory.

4. Dry the leaves to constant dryness (in a drying oven or other warmed place if possible), then weigh them to determine the total organic matter.

5. Average the different sample weights from each trap to determine the organic matter production per square metre per day for each mangrove species measured.

6. During the field visits, observe the mangrove leaves and fragments in the water, as well as any animals that may be feeding on them.

7. Remove the traps at the end of the study.

8. Discuss the importance of mangrove primary productivity. Estimate how much food is produced by the whole mangrove forest by multiplying the production per square metre by the area of the forest.

14. ANIMAL FEEDING

(B, D; a; I, III)

Objectives

1. to observe animal feeding behaviour;
2. to examine what animlas have eaten;
3. to understand how a food chain is determined;
4. to appreciate the relationship between animals important to man and their food supplies.

Curriculum links

Animal behaviour, food chains, herbivores, carnivores, fisheries management.

Preparation

Habitat: any coastal marine habitat where animals can be obtained.

Time required: 2 - 3 hours for each part, divided between field and laboratory activities.

Teacher: select the most appropriate local organisms, preferably macro-herbivores or carnivores; avoid colonial animlas and filter feeders. Try out the exercises in advance to adapt them to local conditions. A familiarity with the anatomy of marine organisms will be useful for the dissections.

Students: introduce concepts of feeding behaviour and adaptations in marine animals before the field activity

Equipment: buckets and other collecting gear
dissecting instruments (scapels or razor blades, pins, forceps, etc.)
aquaria (or large glass jars or plastic basins)
hand lens or dissecting microscope (optional)
microbalance (optional)

Strategy

Question: What and how much do animals eat?

1. Visit a coastal area and collect some large benthic animals or catch some fish that are easy to dissect. Note any examples of feeding behaviour observed, and the environment and possible food sources for the animals collected.

2. Remove the stomach contents of the animals and weigh them (if possible) to see how much the animals have eaten.

3. Examine the stomach contents with a hand lens, dissecting microscope and/or compound microscope to try to identify what the animals have eaten. Compare the fragments observed with what was seen in the animal's environment.

4. Look at the animal's mouth parts. Are they adapted for sucking, biting, scraping, filtering, or catching and swallowing whole?

5. Put an herbivorous animal in an aquarium in conditions that resemble its natural habitat. Be sure that the aquarium conditions will not affect the animal's feeding behaviour. Select a plant that is a known food of the animal. Choose one or more pieces that are easy to quantify. Weigh them, trace their outline if they are flat, or draw them to record the initial amount of plant material. Put them into the aquarium with the animal. After 1 or 2 days reweigh or retrace the plant pieces to see how much has been eaten. Is there obvious evidence of grazing damage?

6. Discuss with the class the significance of their observations. If the animals studied are important in local fisheries, how can a knowledge of their food supply help in fisheries management?

Teaching hints

If it is difficult to obtain animals on the shore, fish for dissection might be obtainable (uncleaned) from a local fisherman.

Ingested fragments of animals and plants are often hard to identify. Spend some time examining the gut contents of local organisms before introducing this as a class project to prevent more questions being raised than can be answeered at the time.

Follow up

Feeding studies can be related to the functional morphologies involved in feeding (See Activity 24, Feeding Adaptations, page 129)

15. FEEDING PREFERENCES

(B, D; III)

Objectives

1. to observe that marine animals have feeding preferences;
2. to learn to test observations experimentally.

Curriculum links

Animal behaviour, feeding, herbivores, animal-plant interactions.

Preparation

Habitat: any coastal area with hard substrate where herbivorous molluscs or similar animals are comon.

Time required: 1 - 2 hours in the field, plus observations in the laboratory or classroom.

Teacher: select a common local marine snail or similar herbivore that is easy to keep several days in an aquarium. Test the organism selected to be sure it will feed in the aquarium befope doing the class experiment. Note that some high-intertidal snails may prefer to be wet but not covered by water.

Students: previous work on other aspects of animal feeding is advisable.

Equipment: marine aquarium
field collecting materials (bucket, knife, etc.)

Strategy

Question: Do marine animals have feeding preferences?

1. Visit a coastal intertidal area where the mollusc selected is common. Observe all the potential food plants available in its habitat.

2. Note where the mollusc is found. Does it occur on or near some plants more than others? Are there signs of feeding on certain plants?

3. If the molluscs are inactive because the surface is dry, try wetting them with seawater. Do they begin feeding after a few minutes wetted?

4. Collect a number of molluscs and samples of the different available food plants and surfaces in their habitat. Return them carefully to the laboratory.

5. Put the molluscs in a marine aquarium with samples of the available food plants attached to rocks or weighed down to make them accessible. Observe them over a few days and record on which plants the molluscs spend the most time feeding. Note that some animals may only feed at certain times of the day or tide. List the plants in order of feeding preference.

6. Discuss with the class the possible explanations for any feeding preferences observed (plant form, texture, taste, etc.) and the significance of feeding preferences for the ecology of the coastal zone. Also discuss possible effects of the laboratory situation as compared to what might occur normally in the field. How might such differences be identified and quantified?

16. MARINE PLANKTON CULTURE*

(B; b; I)

Objectives

1. to observe the role of plankton as food in aquaculture.
2. to learn how to culture phytoplankton.
3. to understand the usefulness of phytoplankton.
4. to develop skills useful for culturing other microorganisms.

Curriculum links

Micro-organisms, nutrition, aquaculture.

Preparation

Habitat:	field excursion to any aquaculture operation or marine laboratory engaged in breeding aquaculture organisms.
Time required:	2 - 3 hours field trip, 2 - 3 hour initial laboratory preparation, and daily observations over 10 days.
Teacher:	obtain phytoplankton samples from an aquaculture farm or marine laboratory and establish stock cultures.
Students:	an introduction to food chains and aquaculture would be useful. Divide students into small groups.

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*Based on an activity devised by Dr. Twee Harmchong.

Equipment: inexpensive aquaria, glass jars or widemouth bottles
plankton net
phytoplankton: Chlorella, chlamydomonas, Tetraselmis, Dunaliella, etc.
disinfectant: formaldehyde, malachite green, etc.
pond water
fertiliser: ammonium sulphate, composted manure, or sterilised soil
microscope slides and cover slips
eye dropper
small air pump (optional)
compound microscope (optional)

Strategy

Question: What do aquaculture organisms eat and how is their food raised?

1. Coastal aquaculture of marine and brackish water animals (prawns, shrimp, clams, oysters, fishes) is spreading rapidly in many countries. The larvae of these marine animals generally eat living phytoplankton (microalgae), which must be cultured in quantity if such aquaculture is to succeed.

2. Students should first visit an aquaculture farm or marine laboratory where they can observe phytoplankton cultures and learn their importance to the organisms being raised.

3. In the classroom or laboratory, each group should prepare a clean aquarium or culture bottle.

4. Disinfect pond water by heating to boiling or by chemical treatment: formaldehyde (37%) 50 ppm for two days, or malachite green 100 ppm for 6 hours. Fill the culture container about $\frac{2}{3}$ full with disenfected water.

5. Add fertiliser: 20 g ammonium sulphate, sterilised soil or sterilised composted manure (approximately 1 kg).

6. Innoculate (add to) the culture container 5 ml of pure stock culture of the phytoplankton.

7. Supply air to the culture container with a small air pump, if available.

8. Place the culture containers in sunlight.

9. Observe the multiplication of the phytoplankton daily for 10 days by recording the colour of the culture water, and checking and recording the density of microalgae under a microscope.

10. Filter the culture at the end of the experiment with the plankton net and observe the quantity of algae produced.

Teaching hints

It is important to avoid any contamination of the phytoplankton cultures by zooplankton.

Be careful that no toxic chemicals or residues get into the cultures.

Phytoplankton cultures can be maintained by harvesting samples and innoculating them into new culture medium.

Follow up

Suggested questions for discussion:

1. What was the difference in density of microalgae between the first and the tenth day?
2. Why did the culture water change colour?
3. What does phytoplankton look like?
4. How many microalgae did you obtain in one litre?
5. Do you believe that microalgae or phytoplankton could be used as human food?
6. What are the reasons for the different precautions and additives?
7. What are the possible sources of any problems with the cultures?

17. CORAL GROWTH RATES

(C; d, a; II, III)

Objectives

1. to observe rates of growth and skeletal accumulation in corals.
2. to learn the importance of precise measurement and data recording in the field.
3. to see species differences.
4. to appreciate the importance of avoiding damage to corals.

Curriculum links

Coral reefs, calcification, growth rates.

Preparation

Habitat	select a field site where different types of corals are common and where access at low tide is not too difficult. A reef flat or shallow back reef area would be suitable.
Time required:	3 - 4 hours for the first visit and 1 - 2 hours for each subsequent visit. Measurements should be made approximately monthly for at least 3 - 4 months and preferably longer.
Teacher:	if class time is limited, the study area can be marked and the corals to be measured can be identified in advance.
Students:	to be able to recognise different types of corals. Some practice in labelling and measuring techniques using dried corals in the classroom may be useful before the field exercise, as precise measurements are required.
Equipment:	plastic rulers (20 or 30 cm long, graduated in mm) cloth measuring tapes (such as used by tailors) string or fishline small rigid plastic tags with punched holes, numbered with pencil or waterproof ink (may be cut out of think plastic food containers) 2 steel rods (reinforcing steel) about 1 m long heavy hammer field notebooks or slates and pencils facemasks or goggles (optional)

Strategy

Question: How fast do corals grow?

1. The rate of coral growth helps determine the rate of reef growth as well as the speed with which a coral reef can recover from damage. This activity involves measuring different corals to see how fast they grow.

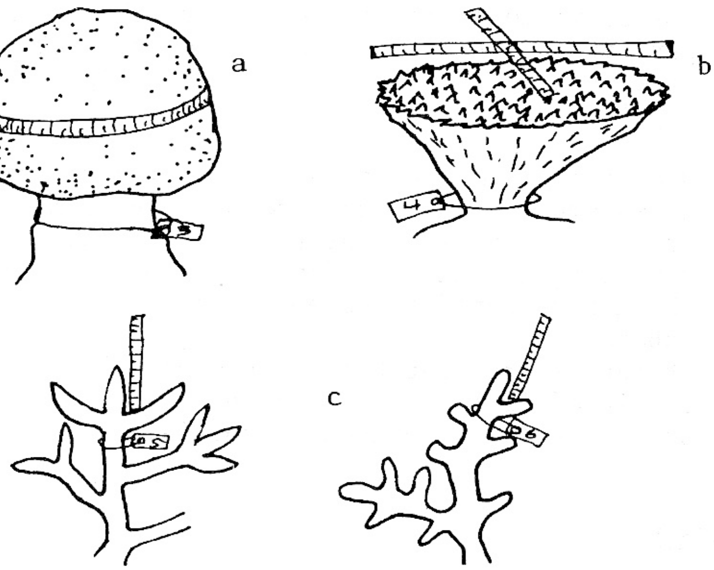
2. The study area should be marked by driving steel rods into the reef so that it can be found again easily.

3. Working singly or in small groups, pupils should select at least one massive or table coral and two types of branching corals tomeasure. The massive or table coral should be of moderate size (20 - 40 cm in diameter) and as close to round as possible. One branching coral should be of the staghorm type (*Acropora cervicornis*, *A. formosa*, etc.) known to grow rapidly.

4. Each coral should be labelled by tying a plastic label firmly around the base (massive/table corals), or loosely around a branch just below the ultimate branch. (Figure 38, page 113)

FIGURE 38

LABELLING AND MEASURING CORALS FOR GROWTH



5. A sketch map should be made showing the locations and numbers of the labelled corals related to the markers.

6. The size of the corals is measured as follows:

a	massive coral:	measure the largest circumference horizontally with a cloth tape measure;
b	table coral:	measure the largest and smallest diameters across the top with a cloth tape measure or ruler;
c	branching coral:	measure the length of the tips from the point of branching just above the label (if there is more than one tip, measure all the tips. (Figure 38)

7. These measurements should be recorded in a field notebook together with the date, the label number, and a sketch of each coral showing what it looks like and how it was measured.

8. The corals should be remeasured in exactly the same way approximately once a month for at least 3 or 4 months. If new branches form on the branching corals this should be noted in the sketches, but measurements should continue to be made from the original starting point.

9. The amount of growth of each coral in circumference, diameter or length should be calculated by subtracting the first from the last measurements. Monthly growth can also be plotted as a graph. The class results for each species can be analysed for statistical significance if desired.

Teaching hints

1. Care must be taken not to break or injure the corals while labelling or measuring them.

2. Since corals grow slowly, considerable precision will be needed in measuring to detect growth. Massive corals in particular may show little or no measurable growth in the time available. This in itself is signfiicant for the pupils to understand.

3. Unexpected results such as decreasing rather than increasing measurements should be discussed in terms of possible sources of error such as damage to the coral or variability in the measuring technique. The amount of variability can be tested through repeated measurements of dried corals in the laboratory.
4. If labels are lost they should only be replaced and measurements continued if there is no doubt as to their precise position, particularly for branching corals.

Follow up

Coral growth rates can be discussed in the class with reference to the size and thus the age of corals observed on the reef, and the rates of coral regeneration after damage by destructive fishing techniques, etc. They can also be related to the rate of growth of the reef itself, and the time it has taken for some reefs to become hundreds of metres thick.

18. ANIMAL POPULATION SIZES

(A, C; a, c; II)

Objectives

1. to determine the numbers of some common benthic (= living on the sea floor) marine animals.
2. to observe the relationship between animal populations, environmental factors and human impact.

Curriculum links

Animal populations, fisheries, environmental impact.

Preparation

Habitat: a shallow coral reef is best, but a rocky shore, mangrove area or mud flat with adequate animals can also be used.

Time required: 2 - 3 hours in the field.

Teacher: select a site with as many interesting benthic animals as possible. The activity and its interpretation will have to be modified somewhat depending on the type of habitat chosen.

Students: the types of animals to be counted should be studied before the field activity. (If the students have experience in data processing, the results can be processed on a micro-computer with suitable programmes prepared before the fieldwork.)

Equipment: a rope and stakes, or square or round quadrat frame, for defining areas within which animals are to be counted
shovels and sieves if the animals are in sediments
field notebooks or slates and pencils
micro-computer (optional)

Strategy

Question: How many animals are there per unit of area.

1. Quantitative measures of animal populations per unit area are often useful in ecology. This activity involves conting some obvious or important animals in the coastal marine environment. The animals counted, the techniques used, and the areas sampled will all depend on the tpye of habitat. The following suggestions for some typical habitats will need to be adapted to local conditions.

2. Lay out one or more areas to be sampled:

coral reef: drive a stake into the reef, and define a 50 square metre area in a circle around the stake using a rope 4 metres long with loops at both ends.

sandy/muddy bottom: use one square metre quadrats, digging up and sieving the sediment to a depth of 20 - 30 cm.

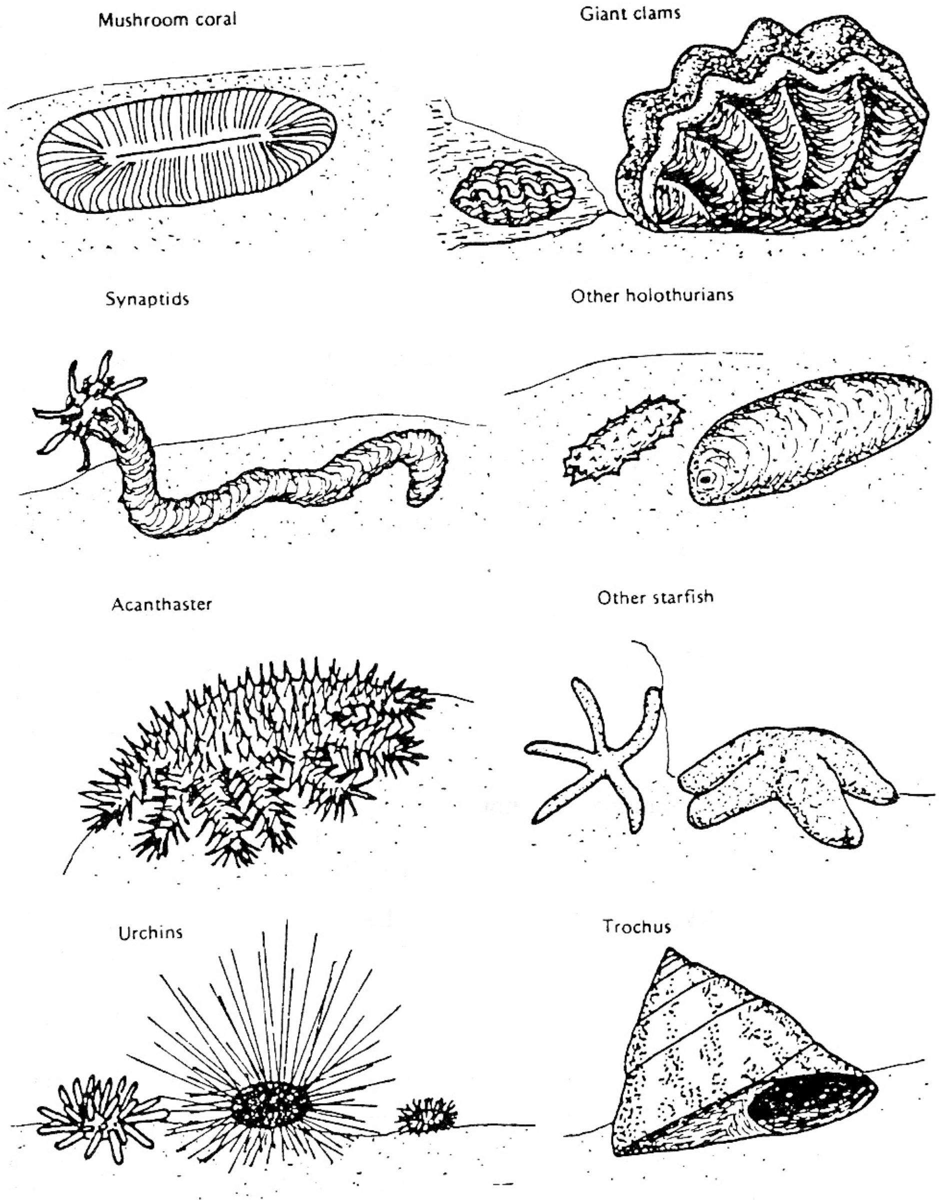
rocky shore: the quadrat size defined by a quadrat frame or rope and stakes can vary from 1 square metre to 50 square metres, depending on the animals to be counted.

3. Count the numbers of common or obvious animals in the sample area, especially those with indicator value:

coral reef: mushroom corals (Fungia spp.) are among the first things to be picked up by tourists; giant clams are often subject to heavy fishing pressure; synaptid holothurians may incrase in number with heavy organic pollution; other holothurians are sometimes fished for food as beche-de-mer; Acanthaster (Crown-of-thorns) starfish may become numerous and damage the reef; other starfish are often obvious and colourful; sea urchins may go through big population changes; large shellfish like Trochus are often collected for food or export. (Figure 39)

FIGURE 39

CORAL REEF ANIMALS TO BE COUNTED



sandy/muddy bottom: bivalve molluscs may be important food organisms; crabs may be numerous and ecologically important.

rocky shore: mussels, oysters and snails may be common food organisms; barnacles may be ecologically important; starfish are important predators, etc.

4. If several samples are taken, compare the results between samples as related to zonation, environmental or human factors.

5. Process the data using tests of statistical significance and presentating the results in graphic form. (Use a micro-computer if available.)

Teaching hints

Select the few animals tha best illustrate topics in the curiculum, or that are important in the local economy.

Follow up

Animal counts lend themselves to monitoring and environmental impact studies (Activities 32 and 33 pages 148and 150) and analysis of the effects of overfishing (Activity 30 page 143). They can also be correlated with environmental parameters.

Reference

Based in part on the Coral Reef Monitoring Handbook, United Nations Environment Programme, Reference Methods for Marine Pollution Studies No. 25 (1984), available from UNEP OCA/PAC, P.O. Box 30552, Nairobi, Kenya.

19. SETTLEMENT AND SUCCESSION

(A, c: a; III)

Objectives

1. to observe the colonization of newly-exposed surfaces.
2. to follow the succession of colonising species over time.
3. to test whether settlement and succession vary in different environments.

Curriculum links

Population dynamics, colonization, succession, competition.

Preparation

Habitat: any intertidal or shallow subtidal area without excessive wave action or sedimentation, preferably with abundant benthic life.

Time required: 1 hour in the field to set up the experiment, plus return visits approximately monthly for three months. The materials collected on the return visits will need 1 - 2 hours of study in the laboratory or classroom after each visit.

Teacher: if literature on local fouling organisms is available it will help in the interpretation of class observations. Some experience in the speed of recolonization locally may be needed to adjust the sampling interval to suit local conditions.

Students: an understanding of reproduction, planktonic larval stages, and fouling will reinforce the effectiveness of this unit.

Equipment: concrete blocks, bricks, distinctive rocks or other heavy hard surfaces, brushed clean and well washed; sterilise by boiling or by washing with alcohol if they have been in the marine environment. (3 per small student group)
buckets
dissecting microscope or hand lenses
steel rods, wire or other means of attachment attachment (optional)
compound microscopes (optional alternative)
glass microscope slides (optional)
masking tape or large rubber bands (optional)

Strategy

Question: How does marine life become established on newly available surfaces?

1. Each small group of students should prepare 3 blocks, bricks or other substrates, taking care that they are clean and free of organisms and toxic substances.

2. The blocks are taken to the field site and placed on the bottom. The location of the blocks should be carefully noted so that they can be found again easily. If time allows, sets of blocks can be put in different zones of the intertidal or at different locations.

3. About 1 month later, the class should return to the same site, collect one block from each environment, and bring it back to the laboratory in a bucket.

4. In the laboratory, they should examine the surface of the blocks with a dissecting microscope or hand lens for any newly settled organisms and make notes describing what they see, with the help of any references that may be available.

5. A second block per site is collected and examined about 1 month later, and the third after 3 months. The observations after 1, 2, and 3 months should be compared.

6. If compound microscopes are available, an alternative rapid microfouling study can be done by attaching 4 microscope slides to each brick with masking tape or rubber bands, leaving the centres uncovered. The bricks are put out intertidally or subtidally and one slide is removed each day for 4 days. The slides are transported carefully to the laboratory and examined uner a compound microscope for diatoms and other micro-organisms that have settled on them.

7. The comparative results over time should be discussed in the class. Which organisms appeared first? Did any organisms become less common over time? If so, what replaced them? Were there signs of grazing on the blocks or slides? Where did the settling organisms come from?

8. The class should prepare a diagram or description of settlement and succession on the blocks or slides

9. If blocks or slides were placed at different levels or sites, the patterns of settlement and succession at each site should be compared.

Teaching hints

This activity needs a site where the blocks will not be disturbed by natural or human factors. The blocks should be heavy enough not to be turned over by waves.

If microscope slides are used, a method should be developed for transporting them to the laboratory, preferably still submerged in sea water, while protecting any organisms from being rubbed off the top surface.

20. REEF FISH SURVEY

(A; a, c, d; I)

Objectives

1. to observe the number of fish on a coral reef;
2. to determine the status of reef fish populations.

Curiculum links

Animal populations, coral reef ecology, fishing effects.

Preparation

Habitat:	a well-developed coral reef area, such as a reef slope, back reef or lagoon reef, where students can safely swim while counting fish. Clear water with low turbidity is desirable (visibility at least 5 metres).
Time required:	1 - 2 hours in the field.
Teacher:	identify sites for the fish surveys that are easily adn safely accessible.
Students:	good swimming ability is required for this activity; familiarity with the types of fish to be counted is important.
Equipment	face masks or goggles snorkels and swim fins (desirable) 100 metres of floating polypropylene rope or string field notebook or underwater slate

Strategy

Question: How healthy are the fish populations on the reef?

1. Select an area along the face of the reef or reef slope parallel to the shore, with fish populations typical for the area, and water depths not exceeding 5 metres.

2. Attach one end of the 100 metre rope to a fixed point (coral head, stake or anchor) or have sufficient students holding it to be able to pull a swimmer back to shore. The student making the survey will swim in a straight line for 100metres along the reef by holding the other end of the rope and swimming until it is stretched to its full length. Fish counts will be made for 2.5 metres on either side of the line (width of the area counted: 5 metres; total area: 500 square metres). One count can be made swimming out along the line, and another swimming back to the starting point.

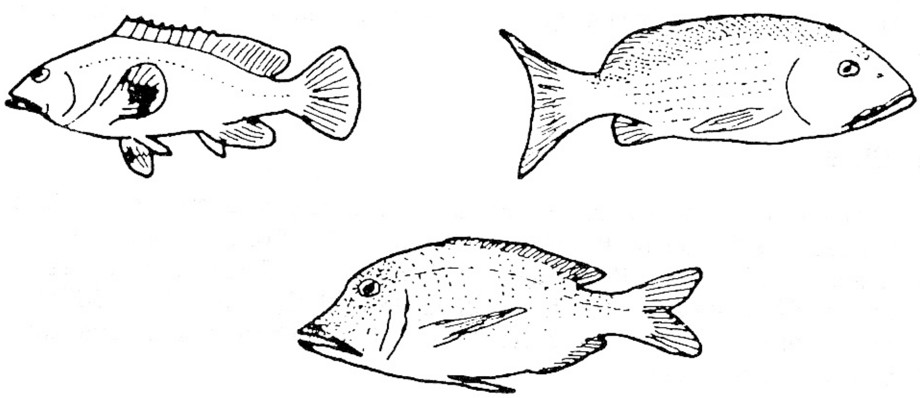
3. Two groups of fish are to be counted (see Figure 40)
Predators: These are larger fish, longer than an outstretched hand, that eat other fish, and include the groupers (Serranidae), the snappers (Lutjanidae) and the emperors (Lethrinidae). (The types of fish expected to be found in a particular area should be ascertained beforehand.) They should be counted first swimming out along the line, as they may be frightened away by a swimmer going by.

Butterfly fish: The butterfly fish or chaetodontids are small brightly coloured reef fish that are quite easy to tell from other fish on the reef. They often swim in pairs and have a special way of biting and sucking around corals. They are not so disturbed by swimmers, tending to stay over the same part of the reef, and so can be counted on the return swim to the starting point.

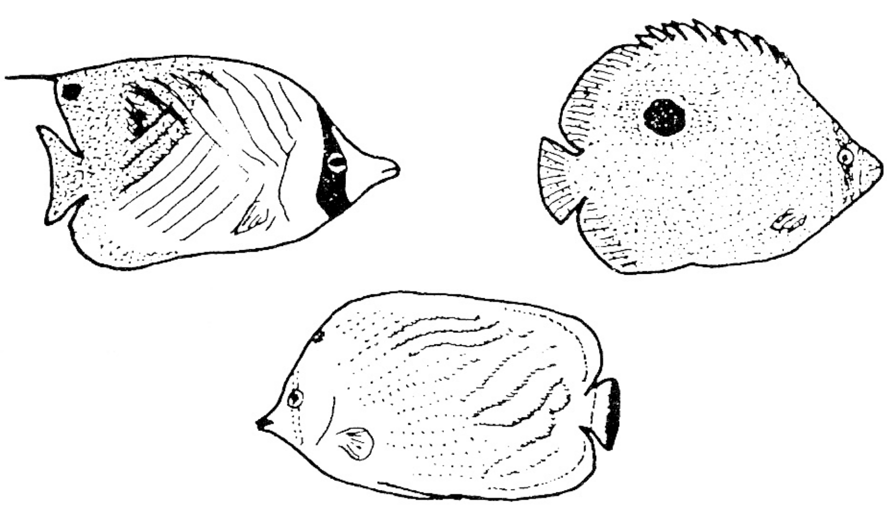
FIGURE 40

REEF FISH TO BE COUNTED

Predators (snappers, groupers, emperors)



Butterfly fish



Fish to be counted (details of shape and colour pattern may vary; not to scale)

4. The swimming student should count the number of predators and the number of butterfly fish in the survey area. Care should be taken not to count the same fish more than once.

5. Other student groups can repeat the survey in adjacent areas for a larger sample of reef fish populations.

6. The significance of the results should be discussed with the class. The numberof larger predatory fish is a good measure of the fishing pressure on the reef. If they are rare or absent, there has probably been overfishing. The small reef fish like the butterfly fish show whether the reef can support many larger fish. Usuallymore than 10 are counted in a 500 square metre survey on a healthy reef. They may decrease as a result of bad fishing methods as when explosives, poisons, small mesh nets or traps are used, or because of aquarium fish collectors. The number of fish can also decrease because something has damaged the coral and other situation that give the fish food and shelter.

Follow up

The reef fish survey method can easily be used to measure overfishing (Activity 30), or environment impact (Activity 33), as well as in monitoring (Activity 32) and conservation surveys (Activity 34).

Reference

This activity is based on a technique described in the Coral Reef Monitoring Handbook, United Nations Environment Programme, Reference Methods for Marine Pollution Studies No. 25 (1984), available from UNEP OCA/PAC, P.O. Box 30552, Nairobi, Kenya. The whole handbook is suitable for use at secondary school level.

21. LOCOMOTION

(A, D; I)

Objectives

1. to observe different types of locomotion in marine animals.
2. to relate functional morphologies to environmental factors.
3. to appreciate the diversity of possible solutions to an evolutionary problem.

Curriculum links

Animal behaviour and adaptations, morphology, evolution.

Prepaation

Habitat: any coastal marine habitat where animals can be observed or collected.

Time required: 2 - 3 hours of field and/or laboratory observations.

Teacher: select the best conditions for observing locomotion in marine animals. If shallow lagoons or tide pools are available, then field observations may be adequate. Otherwise it will be better to catch the animals and put them in aquaria or large glass jars to observe them, either in the field or back in the laboratory.

Students: an introduction to the concept of functional morphology (why organisms have the form they have) is essential.

Equipment: dipnet, knife, scraper, shovel for collecting animals,
glass-bottomed buckets, viewing boxes or face-masks
aquaria or large glass jars with seawater
dissecting microscope (optional)

Strategy

Question: How and why do animals move around?

1. Organisms rarely have features that are not of some use. To appreciate the diversity of life we need to discover why animals are the way they are, and how they have solved the problems of life in their particular environment. This activity looks specifically at some of the kinds of locomotion used by marine animals.

2. Visit an intertidal field site and observe as many animals as possible in their natural environment. Many benthic marine animals move around as larvae, but stay fixed in one place as adults because the moving water brings food to them. Look particularly for those animals that move around as adults. Observe them in the field, and if possible bring some back to watch more closely in the laboratory.

3. Try to observe as many of the following types of animals as possible: fish, starfish, sea urchin or sand dollar, crab, shrimp, an isopod, snail (gastropod), limpet, scallop and worms.

4. For each animal, ask: How does it move around: swimming with fins, a tail or leg paddles; propulsion by squirting water; walking on legs; holding and creeping with muscular foot; pulling with hydraulic feet; pushing or squirming through sand or mud, etc.? Look in detail at the mechanisms for locomotion.

5. Coastal marine animals must be able to move around without being caried away by strong waves or currents. How do the different animals do this: holding on tightly; moving faster than the water, hiding when conditions are bad, etc.?

6. Why does each animal move around in its particular way: to look for food, to escape from predators, to find a mate and reproduce, to avoid bad environmental conditions, etc.?

7. Through class discussion or small working groups, try to define for each animal its strategy for locomotion. For example, the starfish uses hydraulic tube feet to pull itself over the rocks while holding on tightly to avoid being carried away by waves. It seeks food, and can move slowly to escape bad environmental conditions as when it is uncovered at low tide, but it cannot move away quickly from predators.

22. COLORATION

(A, D; I)

Objectives

1. to observe different types of coloration in marine plants and animals.
2. to relate these functional morphologies to environmental factors.
3. to appreciate the diversity of possible solutions to an evolutionary problem.

Curriculum links

Plant and animal adaptations, morphology (form and colour), evolution.

Preparation

Habitat: any coastal marine habitat where plants and animals can be observe or collected.

Time required: 2 - 3 hours of field and/or laboratory observations.

Teacher: select the best conditions for observing the variety of colours in marine plants and animals. Most organisms can be observed in place. For some of the more mobile animals such as fishes, if lagoons or tide pools are available, then field observations may be adequate. Otherwise it will be better to catch them and put them in aquaria or large glass jars to observe them, either in the field or back in the laboratory. However protective coloration will not be as obvious outside of the natural environment.

Students: an introduction to photosynthetic and auxiliary pigments in plants, and to the roles of coloration in animals (why organisms have the colour they have) is necessary.

Equipment: dipnet, knife, scraper, shovel for collecting organisms
glass-bottomed buckets, viewing boxes or facemasks
aquaria or large glass jars with seawater
dissecting microscope (optional)

Strategy

Question: Why are plants and animals coloured the way they are?

1. Even the colours of organisms have a purpose. The many colours of marine plants and animals help them to live better in their environment. This activity looks specifically at the functional roles of coloration used by marine plants and animals.

2. Visit an intertidal field site and observe as many plants and animals as possible in their natural environment. The students should make field notes or drawings of the colours and colour patterns they observe.

3. For the plants, the colours observed should be interpreted in terms of the roles of photosynthetic and auxiliary pigments in absorbing light energy for photosynthesis. Why do marine lants have more such colours than land plants? (Explain in terms of the different spectral composition of light in water.) Is there any evidence of adaptations to protect from too much light (calcification, reflection)?

4. For the animals, look for examples of the following:
   - protective coloration or camouflage to match the background to avoid being seen by predators;
   - colour patterns to permit recognising others of the same species for group behaviour, territoriality or mating;
   - bright obvious colours or patterns to signal that the organism is dangerous or distasteful;
   - patterns or markings to deceive predators as to the size or direction of the organism (location of the head or eye) or to mimic another (dangerous) species.

5. Collect certain animals with protective coloration, and compare their visibility in their natural habitat and in habitats where they do not normally occur. The students should try to put themselves in the position of a predator looking for something to eat.

6. Discuss with the class the selective pressures that could cause such coloration to develop.

Teaching hints

The subject of coloration can first be introduced with classic examples from elsewhere in the world which are often illustrated in books. The students can then look for similar examples in their own environment.

23. PROTECTION

(A, D; I)

Objectives

1. to observe different features providing protection in marine plants and animals.
2. to relate these functional morphologies to environmental factors.
3. to appreciate the diversity of possible solutions to a series of evolutionary problems.

Curriculum links

Form and morphology, adaptation to environmental conditions, animal behaviour, evolution.

Prepaation

Habitat: any coastal intertidal habitat where plants and animals can be observed.

Time required: 2 - 3 hours in the field.

Teacher: prepare in advance a list of the local environmental factors from which intertidal marine organisms must protect themselves. This can also be done as a class exercise.

Students: an introduction to functional morphologies (why organisms have the form they have).

Equipment: dipnet, knife, scraper, shovel for removing or uncovering plants and animals
glass-bottomed buckets, viewing boxes or face-masks

Strategy

Question: How do plants and animals protect themselves in the difficult conditions of the intertidal environment?

1. Organisms must protect themselves from many things in the environment, and many of their features have a protective function. We need to discover how plants and animals have solved the problems of life in their particular environment. This activity looks specifically at some of the kinds of protection used by marine plants and animals.

2. Visit an intertidal field site and observe as many organisms as possible in their natural environment. For each organism, go through the list of factors from which protection is needed, and discuss how the organism has solved the problem of protection. Alternatively, students can observe the organisms and try to decide what their protective features are.

3. Some environmental factors producing protective responses in the tropics are: sunlight, heat, drying out, exposure to rain or freshwater (low salinity), high salinity from evaporation, wave shock and drag in moving water, abrasion by moving sand and rubble, and predation. For each habitat determine which factors are the most apt to be limiting, and how the organisms present protect themselves.

4. Make a list of the kinds of protective features observed. Some common types of protection are: calcified walls and shells (which sometimes can be completely closed or sealed); hard skeletons or cuticles; sharp spines or spicules outside or in the flesh (as in some sponges); flexible joints; tough rubbery texture; water holding capacity inside a shell, between closely packed branches, or inside a sack or spongy tissue; slimy mucus covering; smooth streamlined forms that reduce drag in water; etc. Note that there are oftent rade-offs between different kinds of protection; spines may deter predators but increase drag, a shell may protect from waves but reduce the cooling effect of evaporation. Determine which kinds of protection are most common in different organisms and in different habitats.

<u>Teaching hints</u>

This activity can be organised in different ways depending on the previous studies of the students. Some approaches can be: list all the kinds of protection observed; identify examples of each of the kinds of protection on a list provided by the teacher; describe the kinds of protection used by certain listed plants and animals; define the environmental parameters of certain habitats and identify the protective adaptations of the organisms in them.

<u>Follow up</u>

Experiments can be designed to test the effectiveness of certain protective features, such as the strength of a shell or attachment, the resistance to drying out or to salinity changes, or temperature tolerances (See Activity 26, page 135)

24. FEEDING ADAPTATIONS

(B, D; I)

Objectives

1. to observe different types of feeding adaptations in marine animals.
2. to relate functional morphologies to the constraints imposed by food supply.
3. to appreciate the diversity of possible solutions to an evolutionary problem.

Curriculum links

Feeding, animal behaviour and adaptations, morphology, evolution, symbiosis.

Preparation

Habitat: any coastal marine habitat where animals can be observed or collected.

Time required: 2 - 3 hours of field and/or laboratory observations.

Teacher: select the best conditions for observing feeding activities in marine animals. If shallow lagoons or tide pols are available where animals feed at low tide, then field observations may be adequate. Otherwise it will be better to catch the animals and put them in aquaria or large glass jars to observe them, either in the field or back in the laboratory.

Students: an introduction to types of food and feeding behaviour would be useful before the field activity.

Equipment: dipnet, knife, scraper, shovel for collecting animals
glass-bottomed buckets, viewing boxes or face-masks
aquaria or large glass jars with seawater
dissecting microscope (optional)

Strategy

Question: How do marine animals get their food?

1. All animals have to get food in one way or another. At the same time, the prey species of plants and animals have evolved many defences to aboid being eaten. In the competition to get a share of the limited food available, animals have evolved many ways of feeding. Predators have to find ways of overcoming the defences of their prey, and of specialising or dividing up

the resource to reduce competition. This activity looks specifically at some of the kinds of feeding adaptations used by marine animals.

2. Visit an intertidal field site and observe as many animals as possible in their natural environment. The following categories are some of the feeding adaptations to look for.

3. Some animals simply catch and swallow their prey. They often have large mouths with sharp teeth for holding or taking bites out of prey. Animals that specialise in small prey may have small mouths to match.

4. Other animals sting or paralyse their prey before eating them. They may have many small stinging cells (nematocysts in sea anemones etc.) on their tentacles, or a large poisonous spear or barb. This enables them to eat big or fast animals that would otherwise escape.

5. There are animals, such as snails and sea urchins, that simply scrape or graze their prey off the rock or reef surface. They may have hard beaks for scraping, small teeth or pinchers for nipping or biting, or even a sort of rasp which keeps growing as the end wears out.

6. Others, such as many bivalve molluscus, may simply suck in their food, or even create a flow of water to bring in plankton which they can then filter out.

7. Predators feeding on hard shelled animals can either break the shell, or bore a hole through it, or even slip their stomach through any little space and digest the animals in its own shell.

8. Parasites may simply attach themselves to a host and live on the juices, or even live inside the host's body.

9. Some animals have found the easiest solution of all. They simply grow their food inside their own body, like the corals and giant clams with symbiotic algae; the algae (zooxanthellae) make food from the sun and then release some of it to the host animal in return for fertilisation by some of its waste products. Each one helps the other in what is called a symbiosis.

10. Students can list animals by the types of feeding adaptation they observe, or they can start with a list of feeding adaptations and try to find examples of each type, or be given a list of animals and determine through observation their particular feeding adaptations.

11. Some animals may be easier to observe feeding in an aquarium. Collect both the animals and their preferred food, and place them together where students can observe their feeding behaviour and how their anatomy is adapted to a particular type of feeding.

12. Discuss with the class how different feeding adaptations help to partition the food available in the coastal environment.

25. WATER QUALITY SURVEY

(A, B; c, d; II, IV)

Objectives

1. to survey the water quality a coastal area.
2. to relate the water quality to other observations on the state of the environment.
3. to understand the effects of man on productive ecosystems.

Curriculum links

Water quality, pollution, environmental impact.

Preparation

Habitat: any coastal marine habitat, preferably with both polluted and unpolluted areas.

Time required: 1 or more field trips of 2 - 3 hours, depending on the length of coastline to be surveyed.

Teacher: collect any existing information on coastal water pollution. The sophistication of the survey will depend on the analytical equipment available.

Students: an understanding of the sources and types of water pollution and their effects on the marine environment.

Equipment:
thermometer
secchi disk (20 cm weighted black and white disk attached in its centre to a rope marked in metres)
tape measure (10 m or more)
metre stikc
sieve (2 mm mesh size)
laboratory balance (optional)
device for measuring salinity (hydrometer, refractometer, or conductivity meter; a crude comparative measure of salinity can be made by evaporating known volumes of seawater and the test samples to dryness and weighing).
See page
field noteboosk and pencils

Strategy

Question: What is the quality of the coastal waters?

1. Most water pollution measurements require sophisticated techniques and equipment. However a few simple indicators can be used to determine the general quality of coastal waters. These measures should be made at regular intervals along the coast, perhaps every half kilometre or kilometre, or at least at several points around each harbour, bay, beach or other coastal feature. (See Chapter 4.)

2. At each survey point, take and record the temperature in the shallow inshore waters (5 - 10 cm depth), and if possible in deeper water and offshore. A higher temperature inshore than offshore may be a sign of poor water circulation; a lower temperature may indicate groundwater seepage or other freshwater input.

3. Determine the light penetration in the water with a secchi disc (Figure 26, page 59). Lower the disc from a boat or wharf until it is no longer visible from the surface, and note the depth from the marked rope. Slowly pull it up again until it is just visible, note the depth again, and average the two measurements. This light penetration depth is a good measure of turbidity caused by pollution or suspended sediments.

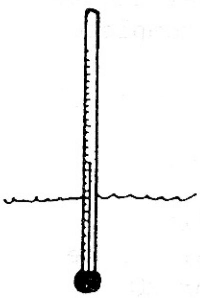
4. Have the students walk along 50 metres of shoreline collecting or counting the number of pieces of plastic or plastic objects observed (bags, bottles, packaging, fishing gear, toys, etc.). Calculate the plastic pollution for each site in terms of the weight of the plastic or the number of plastic objects per metre of shoreline. If plastics are irregularly distributed, it may be necessary to make several measurements and average them.

5. Make a similar count of tar balls in the drift along the shore. On a sandy beach, collect the surface sand and tar balls from a 1 metre wide strip of beach perpendicular to the shore. Wash the sample through a sieve or screen with 2mm mesh, sort out the tar balls by hand, and weigh them or count them and measure their average size (diameter). Take 3 such samples at random on the beach. Calculate the weight (or number) of tar balls per metre of shoreline. Tar balls are a good mesure of nearshore or offshore oil pollution.

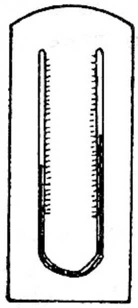
6. If the appropriate equipment is available (Figure 22, page 58), determine the salinity of the coastal water at each site, and compare it with the salinity of seawater from further offshore. If the salinity is lower than seawater, look for signs of freshwater streams or outfalls. If there is no evident sign of freshwater input, there may be groundwater seepage. Such freshwater inputs are important, as they often carry nutrients or pollution from inhabited areas inland.

FIGURE 41

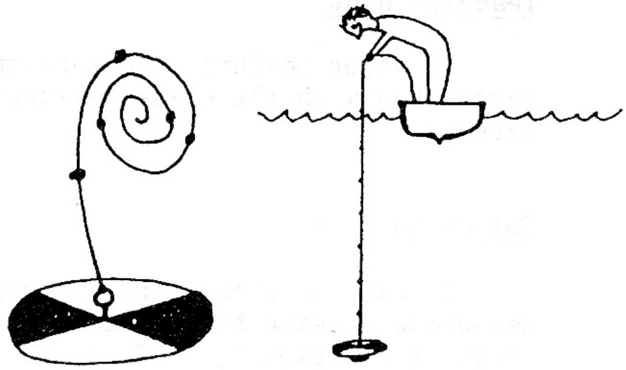
INSTRUMENTS FOR WATER QUALITY ANALYSIS



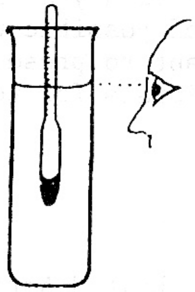
(a) Thermometer



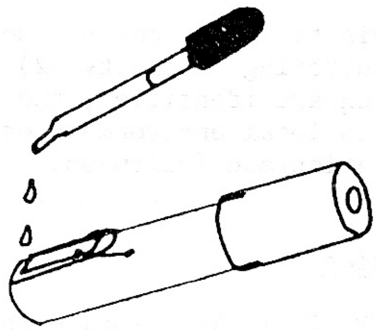
(b) Maximum/minimum thermometer



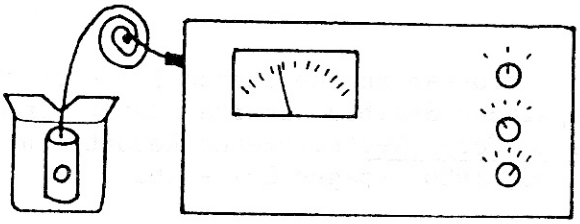
(c) Secchi disc to measure turbidity



(d) Hydrometer



(e) Refractometer



(f) Conductivity meter

7. Prepare a map of the coastline, or other graphic display, and record on it the water quality measurements at each survey point (temperature, turbidity, salinity, plastics and tar balls). Do the signs of water pollution correlate with known activities on the adjacent land?

Teaching hints

If water pollution measurements are available, they can be correlated with the class measurements to make a more complete survey.

Follow up

It can be interesting to compare these water quality measurements with biological measurements along the same shoreline. Activities 1, 2, 3, 5, 7, 12, 13, 17, 18, 19, or 20 would all be appropriate in certain cases, depending on the environment and the effects being studied, to demonstrate the biological effects of a pollution gradient.

This technique can be used by classes each year for long-term monitoring (Activity 32). If signs of possible water pollution are identified, the class may want to present its result to local environment or health officials for possible confirmation and follow-up.

References

SPC/UNEP Coral Reef Monitoring Handbook. United Nations Environment Programme, Reference Methods for Marine Pollution. Studies No. 25, 1984. 25 pp.

UNEP/IOC/IAEA Monitoring of tar on marine beaches. United Nations Environment Programme, Reference Methods in Marine Pollution. Studies No. 15 (draft), 1985. 13 pp.

(Both available from OCA/PAC, UNEP, P.O. Box 30552, Nairobi, Kenya.)

N. G. Willoughby. Man-made flotsam on the strand-lines of the Thousand Islands (Kepulauan Seribu) Jakarta, Java. In Human Induced Damage to Coral Reefs, Unesco Reports in Marine Science. No. 40, 1986. pages 157 - 163.

26. TEMPERATURE TOLERANCE

(A, D; b, c; III)

Objectives

1. to observe the effects of temperature on a marine organism.
2. to determine the temperature tolerance of an organism experimentally.
3. to learn to interpret experimental results in terms of local environmental conditions.

Curriculum links

Temperature, environmental limits, thermal pollution, aquaculture.

Preparation

Habitat: any coastal marine habitat where suitable experimental organisms can be found.

Time required: time for collection of organisms in the field; 2 - 3 hours of laboratory observations; repeat observations after 24 hours.

Teacher: select one or more appropriate common organisms for the experiment. These can be plants or animals, preferably of local ecological or economic significance. If possible the moment of death should be readily observable. Organisms important locally in aquaculture, such as small fish or shrimp, would be ideal. However many marine organisms will not survive in school aquaria, so some preliminary testing will be necessary to find those that will live a week in the laboratory. Intertidal or tide-pool forms may prove most adaptable. Test the experimental set-up to ensure that the organism survives wel in the experimental jars or aquaria before experimenting with temperature variations.

Students: an introduction to temperature effects and temperature tolerances would be useful.

Equipment:
dipnet, knife, scraper, shovel for collecting experimental organisms, and bucket for transport
15 aquaria, beakers or glass jars of about 1 litre capacity
supply of clean seawater
thermometers
maximum-minimum thermometer (optional)
5 large basins or water baths in which 3 aquaria or jars can be placed
ice
source of heat or hot water (or electric aquarium heaters)

Strategy

Question: What is the temperature tolerance of a marine organism and how can it be determined?

1. Every organism has temperatures above and below which it cannot live. This temperature tolerance determines the habitats in which it occurs. Organisms in the intertidal are often exposed to greater temperature extremes than subtidal organisms. Tropical organisms may be less tolerant than temperate organisms, and may be living close to their upper temperature limit.

2. Collect the experimental organism(s) selected and measure the temperature in their natural environment. If possible determine the range of temperatures to which they are normally subjected by looking at temperature records or by leaving a maximum-minimum thermometer in the environment for some time.

3. Transport the organisms rapidly to the laboratory, and hold them for 1 or 2 days to allow them to acclimate to the aquarium conditions.

4. Set up a series of water baths to hold the jars or aquaria. Ideally there should be at least 5 water baths, each able to hold 2 or 3 small jars or aquaria containing several organisms (10 organisms per temperature would be good minimum if possible). One water bath (the control) should be the ambient temperature which should correspond to the temperature in nature. The other water baths should be heated (with hot water) or cooled (with ice) to 5°C and 10°C above and below the ambient temperature. The students should follow these temperatures closely and adjust them as necessary during the experiment. Make graphs of the temperatures to which the organisms are subjected.

5. At the start of the experiment, all the jars should contain organisms at ambient temperature. Place the jars in the water baths, and follow and record the temperature in each jar carefully. Once the temperature in the jar reaches the experimental level (of the water bath) it should be kept there for a half an hour, and then removed back to the ambient temperature. Gentle stirring may help the temperatures to equilibrate more quickly.

6. Notes should be made on the state and behaviour of all the organisms during and after the experiment, as related to the temperatures experienced. Check the state of the organisms again 24 hours later for signs of delayed mortality, before ending the experiment. Note the number of organisms surviving in each experimental condition and in the control. What do the results say about the temperature tolerance of the organism relative to its occurrence in nature? If it is an aquaculture organism, are the results useful in planning aquaculture facilities?

7. If it is possible to have enough experimental organisms and replicate conditions, the results can be tested for statistical significance, or even analysed on a microcomputer.

8. The experiment can be repeated with different organisms to compare temperature tolerances. For instance organisms from tide pools or shallow water might be expected to have higher= temperature tolerances than deeper water forms.

27. POLLUTION EFFECTS

(A; c; III)

Objectives

1. to observe the effects of polluted water on an organism.
2. to learn the principles of toxicity testing.
3. to relate laboratory experiments to field observations.

Curriculum links

Pollution, toxicity, environmental impact.

Preparation

Habitat: any coastal marine habitat where pollution effects are apparent in some areas.

Time required: 2 - 3 hours of field observations and two 2-hour laboratory exercises.

Teacher: select an appropriate experimental marine organism that is common, easy to maintain in laboratory aquaria for a few days, and if possible is absent in known polluted areas but present in environmentally-environmentally-similar unpolluted areas. It can be the same organism as that used for the temperature tolerance test. The moment of death should be readily observable. Identify two coastal areas which are environmentally similar, except that one is polluted and the other is not. The outfall or source of pollution should be evident and accessible if possible.

Students: an introduction to the concept of pollution and pollution effects is necessary.

Equipment: dipnet, knife, scraper, shovel for collecting experimental organisms
field notebooks and pencils
15 aquaria, beakers or glass jars of about 1 litre capacity
supply of clean seawater

Large bottles or buckets for polluted water
graduated cylinderor other volumetric
measure of 1 litre
labels for aquaria
thermometer

<u>Strategy</u>

Question: What is the effect of pollution on a marine organism?

1. Pollution is by definition harmful to at least some organisms. Many kinds of water pollution are toxic and kill more and more organisms as their concentration increases. Polluted waters tend to have fewer kinds of organisms than unpolluted waters. While most techniques for measuring pollutants are too complex for secondary school use, it should be possible to observe the direct effect of polluted water on sensitive organisms.

2. Visit the unpolluted intertidalfield site and observe and record the organisms and communities present. Collect about 50 experimental organisms and allow them to acclimatise to the aquaria in the laboratory.

3. Then visit the polluted site and compare the biological communities with the unpolluted area. Collect a large sample (10 litres) of the polluted water from at or near the source. If the pollution source is of low salinity (freshwater), collect the sample from far enough away that some mixing has occurred, to avoid confusing salinity effects and pollution effects in the experiment.

4. In the laboratory, set up 5 sets of aquaria with three replicates per set. One set should have undiluted polluted water, the next diluted by half with pure seawater to 50%, the third diluted by half again to 25%, and the fourth again diluted 12.5%. The last should have pure seawater as a control. check that the temperature is the same in all aquaria, and that this is close to the temperature in nature.

5. Place some experimental organisms in each aquarium, and observe the immediate effects on behaviour and survival.

6. Check the aquaria again after 24 hours and record the number of surviving test organisms as compared with the controls.

7. Discuss what the results demonstrate about the toxicity of the pollutant. Relate the results to the observed distribution of organisms in the field.

Teaching hints

The experimental details, including the dilutions used, many need to be adjusted to the organism used and the type and concentration of pollutant. Some preliminary trials may be needed to select the organism that best demonstrates the pollution effect. Many pollutants are only released intermittently, so the sample should be taken when the pollutant is known to be present.

In places with little local pollution, this activity can still be done using an artificial pollutant such as copper sulphate.

Follow up

The effect of the pollutant on the environment can be demonstrated more clearly with Activity 33, comparing the polluted and unpolluted areas for environmental impact.

28. FOOD FROM THE SEA

(A; a; I, II)

Objectives

1. to observe what kinds of food are taken from the sea.
2. to measure how much food is taken from the sea.
3. to learn something about fishing techniques and the transport of marketing of seafoods.
4. to understand the importance of the sea to local diets.

Curriculum links

Exploitation of natural resources, fishing.

Preparation

Habitat: any coastal marine habitat where fishing takes place, including if possible a fishing port and fish market.

Time required: one or more visits to observe fishermen's landings and themarketing of fish.

Teacher: obtain any existing statistics on local fish catches. If possible obtain the co-operation of local fishermen in explaining their fishing techniques and showing their catch.

Students: review general information on fishing boats, fishing gear and fishing ports. The principles of harvesting a wild resource, and such measures as catch per unit effort, should be taught together with this activity.

Equipment: field notebooks and pencils
balances or scales for weighing fish
plastic bags or buckets
hand lens or dissecting microscope (optional)
knife
forceps

Strategy

Question: How important is food from the sea locally?

1. Many coastal communities depend on the resources of the sea to provide an important part of the local diet. This activity allows students to measure that importance for themselves.

2. Visit the local landing place as the fishermen come in with their catches, and/or the local fish market. Observe the fishermen separate the marine animals into various groups: economic fish, trash fish, prawns, crabs, etc. Discuss with the fishermen what kind of habitats they fished in and what gear they used.

3. List the names or descriptions of all the different kinds of seafood brought in or available for sale. If some fisheries are seasonal, try to complete the class data from other sources. If possible take examples of the different animals and seaweeds back to the classroom for further examination and identification. (If the collections cannot be worked on immediately, they can be preserved in 10% formaldehyde, and then washed before student use.)

4. Each student should also make a list of the seafood eaten at home over a period of a week. If subsistence fisheries or gleaning by women and children are important, these also should be observed and recorded.

5. If weighing devices are available (the class can make simple balances), quantify the amount of seafood taken in a given time (say 1 week) by weighing fishermen's catches and determining where they came from and how long it took to catch them. Home consumption of seafood can also be weighed over a week.

6. On the basis of the figures collected, make simple estimates of the amount of food produced locally from the sea, perhaps measured per kilometre of coastline, on an annual basis. If the data are available, also determine the fishermen's catch per unit of effort (hours spent fishing per fisherman or per boat).

7. Compare the local catch or consumption of fresh seafood with that of imported seafood (canned, dried or frozen) using import statistics or students' reports of home consumption. Has the proportion of local and imported seafood eaten changed over the years?

8. If there has been a change in seafood consumption, is it due to a change in the local resources (i.e. from over-fishing) or to consumer preferences (better taste, or the ease of opening a can).

9. Discuss the results of the class study in terms of the management of the coastal marine environment.

29. SPAWNING OF BIVALVES*

(D; b; III)

Objectives

1. to learn techniques for inducing spawning in bivalve molluscs.
2. to observe the behaviour of bivalves releasing sperm and eggs.
3. to understand some methods of bivalve aquaculture.

Curriculum links

Reproduction, aquaculture.

Preparation

Habitat: any coastal area where bivalves are present, especially where they are used for aquaculture.

Time required: field trip to collect bivalves, plus classroom exercise for up to 6 - 10 hours or more, depending on the species.

Teacher: Identify common molluscs, especially bivalves accessible on the shore or those used locally for aquaculture, which are appropriate for this activity. Determine the spawning season of the bivalves to select the time when they will be mature and ready to spawn.

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*Based on an activity devised by Dr. Twee Hormchong.

Students: Review concepts of bivalve reproduction, including the anatomy, growth and sexual development of the seed bivalves, and processes of fertilisation and larval development. Understand the economic importance of bivalves and their usefulness in aquaculture.

Equipment:
- small aquaria or big round glass jars.
- heater
- thermometers
- ice
- small plastic bags
- rubber bands
- selected bivalves
- compound miscroscope
- droppers
- microscope slides and cover slips

Strategy

Question: How are bivalves reproduced in large numbers for aquaculture?

1. Bivalves such as oysters, clams and mussels are some of the most important economic marine animals in coastal areas and are widely grown in aquaculture. They are filter feeders that effectively convert plankton into useful animal protein with a high value for domestic consumption and export. Getting them to reproduce at will is an important first step in producing large numbers for aquaculture, followed by raising the larvae until their settlement as young shellfish. This activity shows how to get bivalves to release their eggs and sperm for fertilisation in the seawater.

2. Clean the aquarium or round glass jar. Fill it with seawater which has been sterilised by heating to 50°C for 24 hours or by boiling for 5 minutes, and then cooled to room temperature.

3. Select 4 to 6 bivalves at reproductive maturity, set them in the aquarium or other container, and fill it about $\frac{2}{3}$ full with treated seawater.

4. After 2 hours, put some ice in a small plastic bag, tie it with a rubber band, and put it in the aquarium until the temperature drops to 16 - 18°C. Keep the water at this temperature for 2 hours by adding and removing the ice bag.

5. After 2 hours, add hot seawater little by little to increase the temperature up to 30 - 32°C, and keep this temperature constant for 2 hours.

6. Again add the ice bag to lower the temperature to 16 - 18°C. Continue to alternate between 16 - 18°C and 30 - 32°C at 2 hour intervals until the bivalves release filament-like strands of sperm and eggs.

7. Collect sperm and eggs with an eye dropperm mount them on a microscope. Is it possible to observe fertilisation?

8. Discuss the factors that affect reproduction in molluscs. How long does it take for the fertilised eggs to become larvae?

Teaching hints

Alternative methods for inducing spawning in bivalves are:

- alternating low and high salinity;
- injecting chemicals like hydrogen peroxide (H_2O_2) etc.

30. OVERFISHING

(C; a: I, IV)

Objectives

1. to observe the characteristics of an overfished area.
2. to relate human pressures to their impact on a natural resource.
3. to consider possibilities for the restoration and management of an overused resource.

Curriculum links

Resource use and misuse, fishing, resource management.

Prepaation

Habitat: any coastal marine habitat where overfishing is a problem.

Time required: several 2 - 3 hour periods, depending on methods used.

Teacher: identify a coastal area where overfishing is known to be a problem and fish catches have decreased, and if possible an environmentally similar area where fish catches are normal. Select the activities appropriate to the area that will best demonstrate the effects of overfishing.

Students: prior discussion of the concepts of fisheries management would be useful.

Equipment: see activities referred to in Strategy.

Strategy

Question: What are the effects of overfishing on the environment and how can they be corrected?

1. Overfishing which reduces the productivity of local fisheries is a common problem in many places. Several activities in this manual can be used together to demonstrate the problem and suggest possible solutions. The study can be done just in the overfished area, or preferably by comparing an overfished and an undisturbed or normallyfished area.

2. Using Activity 28 (Food from the Sea), quantify the problem of overfishing, both in terms of catch per kilometre of coastline and of catch per unit of fishing effort.

3. Activity 20 (Reef Fish Survey) can be adapted to the coastal habitat and the most commonly fished species, and used to measure the existing fish populations.

4. One of the activities on zonation and community structure (Activities 2, 3, 4, 5, 7 and 9) can be used to characterise the fishing habitat and possibly to identify environmental changes related to overfishing.

5. Activity 11 (Trophic Levels) can be used to identify important links in the food chain supporting the species fished. Overfishing may affect the balance and structure of caostal food chains.

6. If declining catches may also be related to other factors, a water quality survey (Activity 25) or study of pollution effects (Activity 27) may be appropriate.

7. The overfished area may be compared with an undisturbed area, such as a marine reserve or remote and less frequently visited site, using the environmental impact approach (Activity 33).

8. Discuss the significance of overfishing to local subsistence and the economy, as well as to the productivity of coastal marine ecosystems.

9. On the basis of the data collected fromthe different activities, prepare a list of recommendations to control overfishing and to improve the management of the fisheries resources in the area.

Follow up

The results of the class study and of any recommendations made can be communicated to local fishermen and to fisheries department officials.

An overfished area can also be monitored (Activity 32) to observe changes taking place as a result of fishing or of management efforts to correct the problem.

If the local fishermen agree, a marine reserve (Activity 35) could also be created in part of the area.

31. TRADITIONAL KNOWLEDGE
(A, D; a; IV)

Objectives

1. to collect traditional knowledge about marine resources.
2. to appreciate the value of traditional environmental management.
3. to see how traditional knowledge can help to solve modern problems.

Curiculum links

Environmental management, fishing, traditional cultures.

Preparation

Habitat:	any area where traditional peoples have long-established customs and fishing practices.
Time required:	2 periods of class discussion plus research by students in their own families or villages.
Teacher:	collect whatever information is available on local traditional knowledge of marine resources and practices, such as anthropological or ethnographic studies.
Students:	a review of different methods of fisheries management would be useful background in recognising the value of traditional approaches.
Equipment:	field notebooks cassette recorders for recording interviews (optional) camera for photographing fishing gear and other artifacts (optional)

Strategy

Question: How did the traditional culture use and manage marine resources, and of what value is that knowledge today?

1. The original inhabitants of most coastal areas lived close to the sea and depended on it for survival. They observed nature closely, and over generations came to understand many things about the natural world and how it worked. Most villages had their wise old men and women to whom others would turn for advice and who would decide when and where to fish. Many traditional practices were developed where necessary to ensure adequate food for the people and to protect resources from overuse. In general these systems worked well until the arrival of the Europeans brought new techniques and new economic pressures. The old knowledge and values were often discredited as sorcery or superstition, and children went off to school instead of learning from their elders. Much of this practical knowledge has been lost as old people have died without passing it on to the next generation. Often only fragments are still remembered, or have been recorded by outside observers. Yet much of this information would be useful to the wise management of resources today. It is important to try to save what is left of this traditional knowledge and the value system of respect for nature on which it was often based, so that it can be reinterpreted in the light of modern scientific understanding, and reapplied as necessary to manage resources better.

2. Discuss with the class the significance of local traditional management practices in the marine environment. This could include such things as:

   Fishing methods and materials.
   Knowledge of fish species, their local names, behaviour, migration and reproduction.
   Best fishing locations, times and techniques for each species.
   Controls on fishing: limited access to fishing areas, prohibited (taboo) areas or seasons, catch restrictions, permitted fishing methods.
   Changes in fishing resources, effects of over-fishing, "how things used to be".

3. Students should try to collect as much traditional knowledge of the environment as possible, either locally in their families, or over vacation periods if they return to their home areas or villages. They should ask the elders to recount oral traditions, and to demonstrate traditional ways or making fishing gear. If possible, they should participate in fishing expeditions or other trips where they can learn traditional knowledge first hand.

4. The information collected should be recorded as written notes, or as cassette recordings or photographs if the means to do so are available. If such knowledge is traditionally kept secret within the family, the details need not be shared with the class. What is important is that it is transmitted and appreciated.

5. Another class discussion should be held to report on the results of the student papers or projects.

6. The class can prepare recommendations as to ways in which traditional methods or experience may help to improve the modern management of fisheries resources.

Follow up

The information collected could well be of interest to the local museum or cultural centre. If so, the class can prepare carefully documented archives for deposit in the appropriate institution. The recommendations can also be presented to the elders who co-operated in the projects and to local fishermen.

References

Modified from Training Materials for Rural Enviromental Management prepared by A. L. Dahl for the South Pacific Regional Environment Programme (1984).

For specific examples, see:

Johannes, Robert E. Traditional marine conservation methods in Oceania and their demise. Annual Review of Ecology and Systematics (Palo Alto), Volume 9, 1978, pages 349 -364.

Dahl, Arthur Lyon. Traditional environmental management in New Caledonia: a review of existing knowledge. South Pacific Regional Environment Programme Topic Review (Noumea), No. 18, 1985, 17p.

32. MONITORING

(A, C; a, c, d; II, IV)

Objectives

1. to study stability or change in an environment over time.
2. to observe effects of seasons, weather or population cycles.
3. to learn how to follow the environmental impact of a development project over time.

Curriculum links

Environmental stability and change, seasonality, environmental impact assessemnt.

Preparation

Habitat: any coastal habitat where change may be taking place.

Time required: depends on frequency of monitoring and activities used.

Teacher: choose what is to be monitored locally: seasonality; long-term changes from year to year; the effects of a biological phenomenon such as a population explosion (e.g. Acanthaster Crown-of-thorns starfish) or die-off (e.g. Diadema sea urchins); recovery of the environment after storm damage; or the effects over time of a development project or other environmental change. The technique to be used and its frequency will depend on the type of change and the speed with which it is expected to take place. For rapid short-term changes, the class can make 2 or 3 monitoring surveys during the school year. However most environmental change is gradual, so the activity should be set up with permanent plots where each class survey can be compared with the results of classes in previous years.

Students: the value of repeating a survey to detect changes should be understood. Care must be taken to do exactly the same thing each time the survey is made.

Equipment: same as for the survey technique chosen.

Strategy

Question: What is changing in the environment, if anything?

1. Several activities in this manual describe survey methods that can be used for environmental monitoring. It is simply necessary to repeat the same survey at the same location at regular intervals and to compare the results to see if there has been a change in the environment.

2. The following activities will provide information for monitoring:

   2. Zonation in a sandy-muddy area
   3. The mangrove community
   5. Rocky shore zonation
   6. Zonation on mangrove trunks
   7. Area coverage on a coral reef
   12. Seagrass turnover
   20. Reef fish survey
   25. Water quality survey

   Select the activity and technique that corresponds best to the problem to be monitored. Quantitative techniques will give better results for monitoring than simple qualitative techniques. Greater care in the survey will permit more confidence in the results.

3. Some examples of monitoring activities would be:

   (a) an annual survey to see if long-term changes are taking place in the environment;

   (b) surveys every 4 months around a coastal construction project to see if it is changing the environment;

   (c) surveys every 3 months to see if there are seasonal changes in certain populations;

   (d) surveys every 2 months to observe recovery after severe storm damage;

   (e) surveys each year of a reef affected by Acanthaster to record any decline or recovery of living corals.

4. The area to be monitored should be permanently marked, preferably with several firmly-fixed steel rods or stakes at precisely measured locations, so that each survey is made in exactly the same place. Otherwise sampling error will make it difficult to detect real changes. If one marker is lost, it can usually be replaced by reference to the other markers.

5. Some monitoring activities can be completed with a school year, and the class can see and interpret the results of their efforts. Others may require more than one year to produce results. ach class (except the first) can then compare its results with those obtained in prior years. It is important for the school to keep a permanent record of the results, of the methods used, and of the places studied.

6. The results of the monitoring surveys should be discussed in class, and any changes observed should be related to their possible causes. Some variability may be due to sampling error, some to natural changes, and some to the effects of human activities.

7. If sampling error is suspected, test this by having the class repeat those parts of the survey in the same place at short intervals without looking at the prior results, to see how much variation is due to the survey method.

Teaching hints

Interpreting the results of a monitoring survey is never easy, as many causal factors may be involved. This uncertainty itself should be discussed in class.

A government officer or scientist might be interested in co-operating with the class monitoring survey, and could perhaps explain the results to the class and show their importance for management.

Follow up

Well-documented monitoring surveys can produce results of great interest to fishmen, scientists and government departments responsible for the management of coastal areas and fisheries. The class could prepare a report for submission to the community or to the appropriate authorities.

33. ENVIRONMENTAL IMPACT

(C; a, b, c, d; IV)

Objectives

1. to observe the differences between a disturbed and undisturbed environment.
2. to evaluate the effects of development on the environment.
3. to understand the process of environmental impact assessment.

Curriculum links

Environmental impact assessment, development and environment.

Preparation

Habitat: any coastal marine habitat where impacts of development can be compared with a similar undeveloped or undisturbed area.

Time required: 2 - 3 hours of field work at each of two sites, depending on the methods chosen.

Teacher select two sites which are (or were) as similar as possible in terms of environmental conditions and biological communities, but in which one has suffered the obvious effects of a development project or other impact, and the other is still undisturbed. Decide which activities will best illustrate the nature and extent of the impact.

Students: discuss the different kinds of effects of human activities or development on the environment. The procedures used nationally or internationally for environmental impact assessment can also be reviewed.

Equipment: as required for the activities chosen.

Strategy

Question: What are the environmental effects of development projects and how can they be measured?

1. The assessment of the environmental impact of development projects in order to anticipate and avoid harmful environmental effects is now widely accepted as a standard part of the planning process. Impacts can be measured by studies before and after the development, or by comparing impacted and unimpacted areas. Potential impacts are projected on the basis of the observed effects of similar projects or kinds of disturbance elsewhere. The comparison of impacted and unimpacted areas will allow students both to observe the effects and to experience the process by which impacts are measured or anticipated.

2. Depending on the effects to be observed, choose one or more of the following activities:

   1. Collections of marine life
   2. Zonation in a sandy-muddy area
   3. The mangrove community
   5. Rocky shore zonation
   6. Zonation on mangrove trunks
   7. Area coverage on a coral reef
   9. Coral reef surface area
   11. Trophic levels
   12. Seagrass turnover
   13. Mangrove leaf production
   17. Coral growth rates
   18. Animal population sizes

19. Settlement and succession
20. Reef fish survey
25. Water quality survey
27. Pollution effects
28. Food from the sea.

3. Carry out the same activities both in the impacted area and in the undisturbed area, and compare the results.

4. Discuss and interpret the results in terms of the effects on species and ecosystems of the types of impact demonstrated to have occured in the area.

Follow up

An environmental impact study can be followed by monitoring (Activity 32) to observe if the impact continues or changes over time, or if there is gradual recovery.

34. CONSERVATION SURVEY

(A, D; IV)

Objectives

1. to learn how to make a survey of resources.
2. to determine the priority areas of coastline for conservation action.
3. to understand the importance of conservation as part of development planning.

Curriculum links

Conservation, coastal zone management, planning.

Preparation

Habitat: all the coastline within reach of school field activities.

Time required: 2 - 3 hours of field work per site surveyed; at least 5 sites should be studied, more if the coastline is long or includes many different habitats

Teacher: this activity can integrate information from other activities carried out during the school year, although some additional surveys will almost certainly be required. Class field work can be planned over the whole year with a conservation survey as a final result. The survey can also be made as a separate exercise. Prior to the field work, obtain a good map of the coast, preferably with details of coastal habitats like mangroves and coral reefs. If aerial photographs are available they will also be very useful.

Students: basic concepts of conservation of nature, and of coastal zone planning and management, should be understood.

Equipment: maps and/or aerial photographs of the coastline
large sheets of tracing paper
coloured pens or pencils
field equipment as required by activities selected

Strategy

Question: What areas or ecosystems along the coast have the greatest conservation interest?

1. No country should allow development to destroy all of its natural resources, habitats and ecosystems, which are very important for the future. It is essential that viable examples of each kind of natural system should be conserved, usually in some type of protected area, park or reserve. Coastal ecosystems therefore need to be surveyed for their conservation interest, so that the best or most important examples can be protected for the future. This activity involves identifying the most interesting coastal areas near the school, as a contribution to conservation planning.

2. On the basis of maps, aerial photographs, and personal knowledge of the coast, select a series of survey sites representative of the different types of coastal habitats and features, of spaced at regular intervals along the coast (every 1 to 5 kilometres, depending on the regularity of the coastline). Include any sites known to be of particular interest or biological richness, to have rare or endemic species, or to be critical habitats where organisms come to reproduce.

3. Using the approach described in Activity 1 (Collections of Marine Life) minus the individual collections, prepare a species list for each survey site. Rank the sites according to the number of species found. If any species are of particular rarity or conservation interest, note this separately.

4. Collect information on the ecological interest and richness of each site using those of the following activities that are appropriate to the type of habitat:

   2. Sandy-muddy shore zonation
   3. The mangrove community
   4. Coral reef structure
   5. Rocky shore zonation
   7. Area coverage on a coral reef
   18. Animal population sizes
   20. Reef fish survey

   For each type of habitat (coral reef, mangrove, rocky or sandy shore, lagoon, etc.), determine which are the richest and most interesting ecologically, and rank them in order.

5. Sites with a greater diversity of habitat types should generally rank higher than those with only one type of habitat.

6. If some partsof the coast have been damaged by pollution or development, a water quality survey (Activity 25) at each site will help to show which are the most natural and least disturbed. If old maps or aerial photographs are available, they can be compared with recent information to show what natural areas have already been damaged or lost, particularly to dredging, filling or other coastal construction.

7. Using the different kinds of information collected, the class should select those areas with the greatest interest and importance for conservation, that best represent the coastal resources of their country or region.

8. A large map of the coastline can be prepared showing the distribution of the different coastal habitats, the location of the survey sites, the species numbers, habitat rankings, water quality and other information collected. Areas with the greatest potential for creating marine protected areas can be highlighted.

9. Discuss with the class the importance of a conservation survey as part of comprehensive planning for the management of the coastal zone.

Follow up

Copies of the class conservation survey and map can be presented to local leaders and appropriate govenment officials.

35. A MARINE RESERVE

(C; d; III, IV)

Objectives

1. to create a marine reserve.
2. to observe the effects of protection on species and ecosystems.
3. to appreciate the values of protected areas for resource management.

Curriculum links

Conservation, coastal zone management, fisheries.

Preparation

Habitat: any coastal marine habitat that is already protected or that can be protected as a school reserve.

Time required: 2 - 3 hours of field observations each 1 - 2 months.

Teacher: if a marine protected area already exists near the school, obtain permission from the responsible authorities to study some part of the area. If there is no such area, obtain the approval of local authorities and/or fishermen for the creation of a small school reserve in an accessible coastal area.

Students: background information on conservation and the role of protected areas.

Equipment: signs and markers to designate the protected area field equipment as required for activities selected.

Strategy

Question: What is the value of a protected area?

1. Protected areas such as marine parks and reserves are an important pat of national conservation and development plans. They preserve undisturbed examples of natural ecosystems, and may serve as a refuge and breeding area where species that are fished or collected can multiply and repopulate adjacent exploited areas. If there is no existing marine protected area, select an appropriate site as determined by accessibility to the school and/or conservation interest as determined by a conservation survey (Activity 34). An area approximately 100 - 300 metres on a side should be sufficient for the school project, although a larger area would be of greater value for conservation.

2. Obtain the agreement of local and/or national authorities and local fishermen and land owners that the area will be considered as a school nature reserve, and that fishing and other exploitative activities will not be done in the area, at least for the school year, if not for an indefinite period. The site could be legally set aside as a protected area, but protection may be equally effective if it is by informal agreement with local fishermen and other users.

3. Mark the boundaries of the area with signs and marker poles so that people are reminded of its protected status.

4. Survey the school reserve using the appropriate activities, but avoiding any collections or damaging activities. Survey activities may include:

   2. Sandy-muddy shore zonation
   3. The mangrove community
   4. Coral reef structure
   5. Rocky shore zonation
   7. Area coverage on a coral reef
   18. Animal population size
   20. Reef fish survey.

5. Repeat the survey every 2 - 3 months to observe the effects of protection on the species and communities. Note particularly the status of any species that were previously fished or exploited.

6. If there is a similar but unprotected area not too far away, make comparative surveys to show the differences between the protected and unprotected areas.

7. Surveys of fish (Activity 20) and food from the sea (Activity 28) can be made or repeated in adjacent waters to see if the existence of the reserve had led to better fish populations and catches outside the reserve.

8. Other marine activities can also be carried out in the school reserve as long as they respect its reserve status and do not involve collecting or damaging organisms.

9. Prepare a class report on the reserve and on the effects of its protected status, to be shared with local fishermen and authorities.

Follow up

If the class studies have demonstrated the effectiveness of the school reserve, try to have it established on a permanent basis.

36. MARINE AQUARIUM

(B, D; b, d; III)

Objectives

1. to learn methods of rearing marine fishes in an aquarium.
2. to observe fish behaviour.
3. to understand the complexity of maintaining a suitable environment for marine life.
4. to appreciate the value and beauty of marine aquaria.

Curriculum links

Animal behaviour, effects of physical and biological factors, nutrition, mortality.

Preparation

Habitat: fish may be collected from any coastal environment.

Time required: 2 - 3 hours to set up aquarium, one or more field trips to collect fish (optional), plus regular daily care.

Teacher: obtain and study reference works on rearing marine fishes in an aquarium (often available from pet or hobby shops). Determine the best supply of marine aquarium fishes: commercial purchase, local collectors, or collection by the class at a suitable site. Check on any government regulations concerning aquarium fish collecting. If there is a marine aquarium in the area, arrange for a class visit.

Students: review the principles of marine fish rearing in aquaria. Organise in groups of 4 - 5 to take on the different responsibilities for the aquarium.

Equipment:
inexpensive aquarium of glass, plastic or plexiglass with walls 13 - 19 mm ($\frac{1}{2}$ - $\frac{3}{4}$ in) thick
sea water of 30 ppt salinity and pH 7.9 to 8.5
water filter set (if available)
inexpensive air pump
light (ordinary electric or fluorescent lamp)
dip net
marine aquarium fishes
marine fish food
disenfectant: formaldehyde, copper sulphate, methylene blue, malachite green and/or potassium permanganate
medicines as needed for disease treatment
thermometer
pH test kit or meter
salinity measuring device

Strategy

Question: How can marine fishes be raised in an aquarium?

1. Prepare the aquarium by cleaning it. Purify the sea water by filtering; check that the salinity is about 30 parts per thousand (ppt) and the pH between 7.9 and 8.5. Place a considerable amount of sterilised sand, stone,coral sand or gravel in the bottom of the aquarium. Fill the aquarium about $\frac{2}{3}$ full with treated sea water. Aerate the aquarium with a small air pump. Filter the water constantly to keep it clean, and clean the filter regularly.

2. Obtain mature marine fishes or collect them from a suitable coastal site, disinfect them so that they are free from diseases and parasites, and put them into the prepared aquarium at an average density of 1 - 2 fish per 10 litres (3 - 6 fish per foot^3). For large fish the density should be less.

   Some kinds of marine aquarium fish are:

   - beautiful fishes: butterfly fish, angel fish, parrot fish, red soldier fish, wrasses and batfish;
   - coral fishes or sea anemone fishes: clown fish, pink skunk anemone fish, yellow tailed anemone fish, anemone fish, etc.;
   - strange fishes: trunkfish, pufferfish, triggerfish, spiny puffer, procupine fish, lion fish, eels, rays, sea horses, etc.;
   - economic fishes: giant grouper, grouper, red snapper, sea bass, milk fish, etc.

3. Arrange the lighting according to the kinds of fish and where they are found: clown anemone fish, butterfly fish and angel fish prefer strong light; seahorses, batfish and groupers prefer mediumlight; and giant groupers, eels and lionfish prefer low light.

4. Feed the fish daily with food appropriate to the kinds of fish:

   - fresh food: fish, shrimp and oyster flesh;
   - living food: shrimp larvae, <u>Artemia</u>;
   - planktonic food:
     - phytoplankton: <u>Chlorella</u>, <u>Isochrysis</u>, <u>Chaetoceros</u>;
     - zooplankton: protozoa, rotifers, <u>Artemia</u>;
   - Commerical instant marine fish food.

5. Clean the aquarium after feeding.

6. Keep a close watch for fish diseases and treat them if necessary:

Disease	Treatment
Viral/bacterial diseases	- separate diseased fish - treat with antibiotics
Parasitic diseases:	
- fungus: Saprolegnia	- separate diseased fish - sterilise aquarium with copper sulphate or formaldehyde - treat with antifungal medicine
- dinoflagellate: Oodinium	- same treatment as for fungus
- external protozoa: Costia	- separate diseased fish - disinfect aquarium with formaldehyde, copper sulphate, methylene blue or malachite green
- intradermal protozoa: Ichthyophthirius	- separate diseased fish - sterilise with formaldehyde, malachite green or copper sulphate - raise water termperature as high as fish can endure
- intestinal protozoa: Hexamita	- apply Enheptin
- external trematoda: Gyrodactylus	- immerse in formaldehyde or potassium permangante
- parasitic worms: trematoda, cestodes, nematodes and acanthocephatids	- eradicate hosts or intermediate host such as gastropods or crustaceans and apply Kamala for cestodes and Piperazine citrate for round worms.
- crustanceans: Ergasilus	- pinch with pincers or immerse in potassium permanganate or formaldehyde.

7. Observe and record fish behaviour such as feeding activities, group behaviour, mating and reproduction.

8. Record changes in each fish such as growth, colour, appearance, behaviour, etc.

Teaching hints

Supervise the student activities closely, as one mistake can result in the death of all the fish. Consult a local marine aquarium specialist for details on the preparation of disinfecting solutions and medicines, for the symptoms of the various diseases, and for any other problems that may arise. Check the sea water from time to time for salinity, pH, cleanliness, etc. and take corrective action as necessary.