

### III. BIOLOGICAL MONITORING

---

#### OVERVIEW

In designing a coral reef monitoring program, you'll need to make some compromises between the accuracy and completeness of your data, and the time, difficulty and/or expense of getting it. You may also need to consider whether the risk of damaging the reef by using certain monitoring methods is warranted.

Your monitoring program will need to reflect the diversity of your study area through the study sites selected (composition) and respond to any changes that occur over time. For example, changes in coral cover can be adequately measured with much less sampling effort when cover is uniformly high than when it is low and patchy. No single set of measurements will be ideal or even workable for all locations or at all times, and your methodology must be flexible in order to avoid over or under-sampling.

Since using only one data-gathering technique is unlikely to provide all the information that will be useful to you, it's best to use a combination of methods, if possible. This manual emphasizes documenting changes in percent cover and the spatial arrangement of stony corals (including the fire coral *Millepora*) because they create the structure of the reef. Some procedures for monitoring octocorals, sponges, algae and fish are also included.

Certain marine organisms, often referred to as "keystone species", have functional roles that are more important than their abundance or biomass suggests. Changes in the population size and distribution of these species can be reliable indicators of broader changes in the local marine community. Examples of keystone species are: starfish (*Acanthaster*) in Pacific coral reefs, and sea urchins (*Diadema antillarum*) in the Caribbean.



## Sampling Units

This manual includes three approaches to monitoring corals and other reef components: through the use of individual colonies, quadrats, and linear transects. These approaches may be used separately or combined with each other, and underwater photography is an important complement to any of them. The following chart summarizes the kinds of monitoring each sampling unit is best suited for. For a more detailed comparison of quadrats, photo-quadrats, and chain transects, see page III-12.

Sampling Unit	Monitoring Uses
Coral colony	Monitor general condition of specific stony corals, including growth, bleaching, diseases, algal overgrowth.
Quadrat	Measure percent cover, species diversity, relative abundance, density and size; and monitor corals, octocorals, sponges, seagrasses, and algae.
Linear transect	Measure percent cover, species diversity and relative abundance in zones dominated by head corals; estimate spatial index; unsuited to elkhorn zones, octocoral-dominated pavement areas, or areas where colonies are small and scattered.

## Monitoring Frequency

Monthly observations are generally best for monitoring individual coral colonies. Quadrat and transect surveys done every six months provide sufficient data for assessing changes in percent cover and species diversity, and reduce the risk of damaging reef organisms during the survey process. Of course, in the event of a storm, oil spill or other disturbance, it's important to assess the effects as soon as possible, survey permanent quadrats or transects for which data were obtained before the disturbance, and continue to monitor the aftermath and recovery.

## Data Analysis

It's easy to collect more data than you have the time, resources, or budget to analyze immediately, but don't allow raw data to accumulate to the point where analysis becomes overwhelming or occurs too late to be useful. Short-term data can be helpful in getting financial support for long-term monitoring goals. In planning your monitoring program, be sure to carefully gauge the effort and expense that may be involved in analysis.

### Reference

Hughes, T.P. (1992) "Monitoring of coral reefs: a bandwagon?" *Reef Encounter* 11:9-12

## MONITORING INDIVIDUAL STONY CORAL COLONIES

### General Condition

Observing individual stony (hard) coral colonies over time can be a simple way to monitor bleaching, algal overgrowth, predation, disease, sediment smothering, and damage from SCUBA divers or snorkelers. Here are the basic steps for gathering baseline data.

- 1) **Mark the colony.** The colony can be marked for future monitoring by using a plastic cable tie with number-coded “cattle tag” attached to a 3-inch hardened masonry nail that has been driven into the substrate near the colony. Any encrusting organisms that grow on the tags can be scraped off to reveal the number.

As each colony is marked, record the compass bearing and distance from the previous colony, and which side of the colony the tag is on (N,S,E,W). It’s also a good idea to place a few survey stakes or other reference markers nearby and record the distance and compass bearings to each colony or group of colonies. If the coral colonies are dislodged by a storm, the reference markers are more likely to survive and enable you to locate the remaining marked colonies.



Marked Colony

- 2) **Identify the species.** Over 40 species of stony corals appear in the Caribbean and western Atlantic. Learning to identify them takes practice and a good reference book. (See “Information Sources,” VI-S, for suggestions.) The taxonomy of corals, like that of many animals, is subject to change. For example, it’s been suggested that *Montastraea annularis*, long considered the most abundant and wide-ranging coral in the region, is actually three “sibling” species, two of which have significantly different growth rates, and one of which has unusual coloration that may be confused with bleaching

### Reference

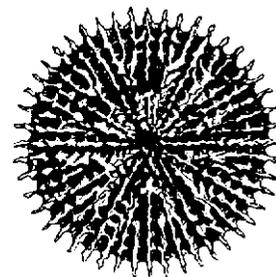
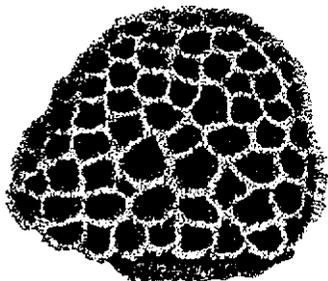
Knowlton, N., Weil, E., Weigt, L., Guzman, H. (1992) “Sibling species in *Montastraea annularis*, coral bleaching, and the coral climate record,” *Science* 255:300-333.

3) Record the condition. The suggested abbreviations shown below may be helpful in describing the colony.

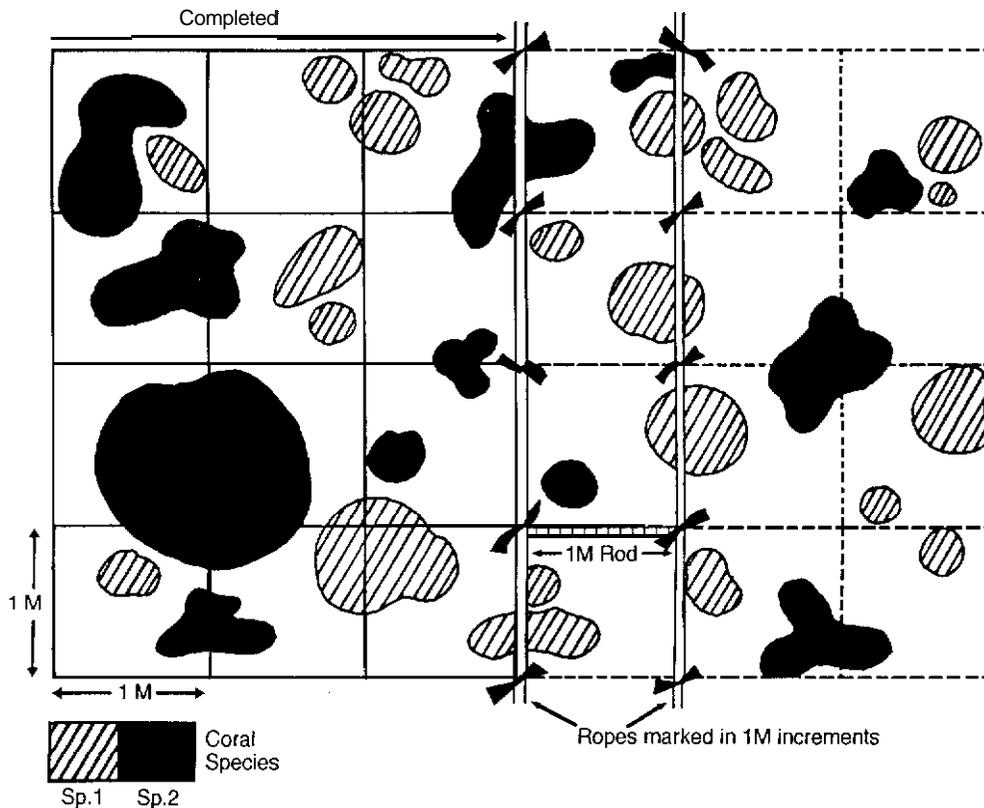
Abbrev.	Condition	Abbrev.	Condition
blea	bleached coral; white, with tissue remaining	fr	freshly grazed coral tissue
dcw	dead coral; white, cleaned coral skeleton without tissue	gr	older grazed area, overgrown with algae
dcs\turf	cleaned coral skeleton without tissue but with algal turf grown over the skeleton	rg	older grazed area that has been grown-over by tissue
dca	dead coral with algal turf; skeleton not visible or conspicuous, older	bbd	black band disease
light	light-colored; bleached but not completely white	wbd	white band disease
lr	light ridges (noted on <i>D. strigosa</i> )	mucus	mucus coat, noted on <i>P. astreoides</i> ; often with adhering sediment or algal growth; <i>S. siderea</i> has tufts or globs of adhering mucus; often seen in summer in Florida
discol	discolored tissue: unusual color not due to bleaching -- purple, pink, bluish, etc. ( <i>S. siderea</i> has a violet color which appears luminescent during bleaching events)	mustard	mustard-colored tissue on <i>P. astreoides</i>
		ss	sediment spot; small sediment-filled spot or hole

4) Photograph the colony. Each colony should be photographed or videotaped when its condition is first recorded. Include a slate in the photo with an identification number, date and scale (a color chart in the photo may help document subtle changes over time). Take the photo from the angle which best represents the entire colony.

You can have a photograph laminated and bring it with you in the next survey to assess changes. These photographs are intended for qualitative rather than quantitative analysis because it's impossible to replicate the camera angle and distance exactly unless a rigid framer is used. For more information about underwater photography, see III-29.



**Mapping the colonies:** If you are tagging colonies that are abundant within a limited area (up to 100m<sup>2</sup>), you may find it useful to create a map. Draw the coral colonies to scale on a grid while underwater. To position the colonies on the grid (each square equals 1m<sup>2</sup>), lay two weighted lines (marked in meters with flagging tape) parallel to each other, one meter apart. Leap-frog one line over the other until you have mapped the entire area. Maintain a compass bearing along both weighted lines to keep them parallel. Although this technique provides a good way to relocate colonies for repeat sampling and document their condition over time, it is labor intensive: a 100m<sup>2</sup> area at 3-4 m depth can take 6 hours to map.



**Map of Coral Colonies**

**Monitoring frequency:** Monthly observations are generally the most effective for assessing the cause and effect of changes in individual stony coral colonies. If observations are done less often, damage that has occurred since the last survey may be difficult to see. Algae may grow rapidly over freshly broken areas, sometimes within a week.

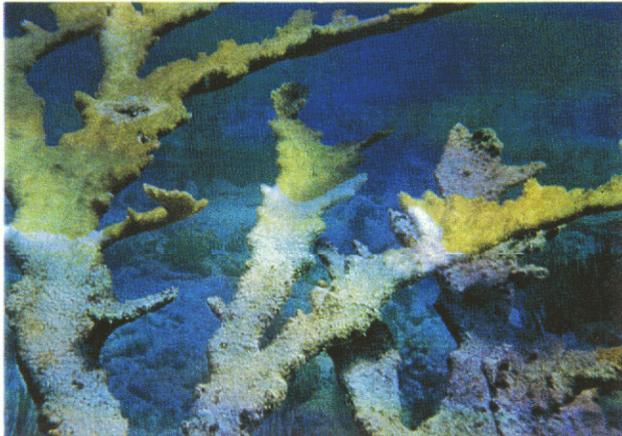
References

French, K., Lewis, S., Hillis, Z. (1990) Coral monitoring conditions guide at the Buck Island Reef National Monument, report prepared for the U.S. Department of the Interior, National Park Service.

Bythell, J.C. Gladfelter, E.H. and M. Bythell (1993) "Chronic and catastrophic natural mortality of three common Caribbean reef corals," *Coral Reef* 12(3/4): 143-152.

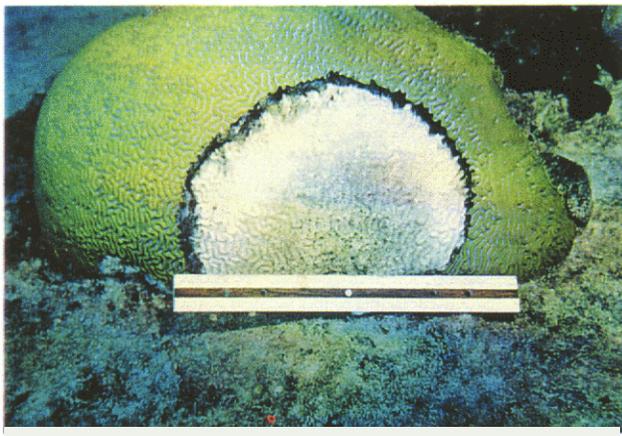
## Coral Diseases

Diseases of stony corals have been observed throughout the Caribbean and western Atlantic, even on reefs far removed from human activities. Monitoring coral colonies will provide information on the distribution of diseases throughout the region. We need to learn more about the extent and severity of these diseases and their causes. To assess the effects of coral diseases, it is best to monitor individual colonies.



*Elkhorn with White Band Disease*

White **Band; Of** unknown etiology; primarily affects species of *Acropora*. It appears as a white band that begins around the base of a colony or branch and progresses distally. The "white band" is bare skeleton devoid of zooxanthellae-bearing coral tissue. White band disease usually progresses rapidly, killing the entire colony.



*Head Coral with Black Band Disease*

**Black Band:** Caused by the cyanobacterium ("blue-green alga") *Phormidium coralyticum*, most commonly infecting *Montastraea annularis*, *Diploria spp.*, *Colpophyllia natans*, and *Siderastrea siderea*. It shows up as a dark pencil-thick band that forms a halo on the coral head. The area on one side of the band appears normal, while the other side exhibits a gradual shift from stark-white to algal-covered skeleton. The coral may die from the disease or it may eventually grow back over the dead area.

## References

- Gladfelter, W.B. (1982) "White-band disease in *Acropora palmata*: implications for the structure and growth of shallow reefs," *Bulletin of Marine Science* 32(2):639-643.
- Rutzler, K., Santavy, D. (1983) "The black band disease of Atlantic reef corals; I. Description of the cyanophyte pathogen," *PSZNI Marine Ecology* 4(4):301-319.

## Coral Bleaching

The tissues of stony corals and other symbiont-bearing Cnidarians appear bleached when they lose their endosymbiotic algae (zooxanthellae), or when the zooxanthellae lose their pigmentation. Bleaching must be distinguished from "photo-acclimation", in which the concentration of pigments changes simply in response to changes in light. There is some evidence that bleaching may be a response to higher water temperatures and/or ultraviolet radiation. Significant bleaching events occurred in the Caribbean in 1987 and 1990. To monitor bleaching activities, it is best to collect data on individual colonies.

In Florida, the rooanthid known as the golden sea mat (*Palythoa*), which has been observed as the first organism to exhibit bleaching from increased water temperature, may be used as an Indicator species for bleaching. It usually precedes bleaching of *Montastraea* by a week. Mortality of part or all of a bleached coral can occur and algae may grow over the stark white skeleton. Some species of Octocorallia (e.g., *Briareum asbestinum*, *Plexaura homomalla*), and sponges may also appear bleached due to loss of their symbiotic algae.



When bleaching or white band disease is suspected, it's important to consider other possible causes for changes in appearance, such as:

- predation by the "tireworm" *Hermodice sp.* or the mollusk *Coralliophila sp.*, both of which can leave large areas of stark white skeleton.
- a nearby sea fan (octocoral) or clump of macroalgae (such as *Dictyota*) that shades or abrades the coral.
- the use of coral heads as handholds in SCUBA-diving areas causing death and eventual coverage by algae.
- color differences in coral species between areas and individuals; e.g., the purple *Porites sp.* of St. John, USVI has never been seen at Buck Island, St. Croix, USVI, which is only 35 miles away.

---

## How to Monitor Bleached Corals

- 1) Record the following information:
  - ▶ when and where the bleaching was first noticed
  - ▶ which species are affected
  - ▶ the size range of the colonies
  - ▶ which parts are bleached or pale (e.g., ridges, branch tips, grooves)
  - ▶ the depth at which affected colonies are growing
  - ▶ the density of bleached colonies, i.e., the number within a known area (e.g., 12 colonies in a 10m<sup>2</sup> area)
  - ▶ any unusual environmental conditions (storms, oil spills)
- 2) Tag both bleached and unbleached colonies with numbered tags, following the procedures outlined above under "General Condition."
- 3) Photograph the colonies and, if possible, videotape them. Photography dependent on natural light will be affected by depth, light quality in the water column, time of day and season. If you are evaluating color quantitatively, use a strobe, the same type of film, and the same F stop for all photographs. Including a color chart in the photo which has gradations of the color characteristic of the species can help document subtle changes.
- 4) To assess recovery and/or condition over time, observe and photograph each study coral every one to two months and record changes. Are polyps extended? Is mucus being released?
- 5) Select a subset of colonies for growth measurements using one of the methods described in the next section, "Coral Growth".
- 6) Record data on temperature, turbidity, salinity, and light if possible.

i...

If adequate laboratory facilities are available, you can sample tissue from affected colonies and centrifuge it to determine both the density of the zooxanthellae and the concentration of pigment.

### References

D'Elia, C.F., Buddemeier, R.W., Smith, S.V. (1991) "Workshop on Coral Bleaching, Coral Reef Ecosystems and Global Change," Report of Proceedings, Maryland Sea Grant College Publication, College Park, Maryland.

Jaap, W.C. (1985) "An epidemic zooxanthellae expulsion during 1983 in the lower Florida Keys coral reefs: hyperthermic etiology," Proceedings of the 5th International Coral Reef Symposium, 6:143-148.

## Coral Growth

The growth rate of a coral colony is highly variable. It depends on the species and may fluctuate significantly within an individual colony and from month to month. While *M. annularis* may grow less than 1 cm a year, branching corals such as *A. palmata* may grow as much as 10 centimeters a year. Four ways to monitor coral growth are described below, from the simplest to the most complicated. The method you choose may depend on what kind of coral you are monitoring.

**Branching corals:** To provide a baseline for measuring growth in branching corals, you can wrap a plastic cable tie around a branch with a tag to identify the sample. Using a flexible plastic ruler, periodically measure the distance from the baseline to the end of the branch to determine the net linear extension. Growth may be evident in measurements taken as often as once a month. Note that over time the cable tie may become embedded in the skeleton.



*Branching Coral with Cable Tie*

**Head coral:** A simple way to monitor growth in head corals is to drive a nail into the substrate next to the head, or into a dead portion of the coral. Tag the nail and use it as a reference point from which to make measurements.

**Photographs:** With relatively flat head and plate corals, you can estimate growth by digitizing the photographic image to calculate the area of living coral and comparing data from successive photographs. Growth may be evident in measurements taken every six months. However, make sure to take the photos from the exact same angle and distance each time, and to place a scale (such as a ruler) along the edge of the photo-quadrat for referencing size. For more guidance, see "Underwater Photography", III-29.

*Alizarin Red S Bone Stain*: If done correctly, this method can provide a more precise measurement of coral growth than photographs, and it can be used with either branching or head corals. However, it requires removing live coral and does not provide measurement of the same coral's growth at subsequent intervals.

### Measuring Coral Growth with Alizarin Red S Bone Stain

- 1) Place a clear plastic 1-liter bag containing about 20 mg of the stain over the end of a coral branch or over an entire colony and tie it off in one corner securely with cotton string. The final concentration of stain in the bag will be about 10-15 mg/l seawater. If the stain is too diluted, it will be difficult to measure the growth.
- 2) Remove the bags after 24 hours except in the case of *A. palmata*. Four hours is sufficient for staining faster-growing corals like *A. palmata* but not for slower growing head corals. Also, in high-energy zones where *A. palmata* often grows, wave and surge action can move the plastic bags back and forth, damaging the coral colony.
- 3) Label each treated coral with a numbered plastic tag.
- 4) Collect the corals after 2 to 4 months, remove the live coral tissue with a water jet, and measure the growth since staining.

. For *Acroporu* and other branching species:

To determine the linear extension of new skeleton, take the mean of 10 equally spaced measurements along the zone of the new growth.

To determine the calcification rate per branch perimeter, remove the new (unstained) growth with a saw and divide its weight by the perimeter of the branch at the base of the new growth and the number of days since staining. This value (g/cm/time) relates calcification to the original amount of calcifying tissue present.

► For head corals:

To determine the growth of roughly hemispherical corals, section them vertically through the center with a rock saw. Measure the distance between the upper limit of the stain line and the periphery of the colony using a dissecting microscope and ocular micrometer. The average of 10 such measurements spaced evenly across the colony can be used as a mean extension value for that colony.

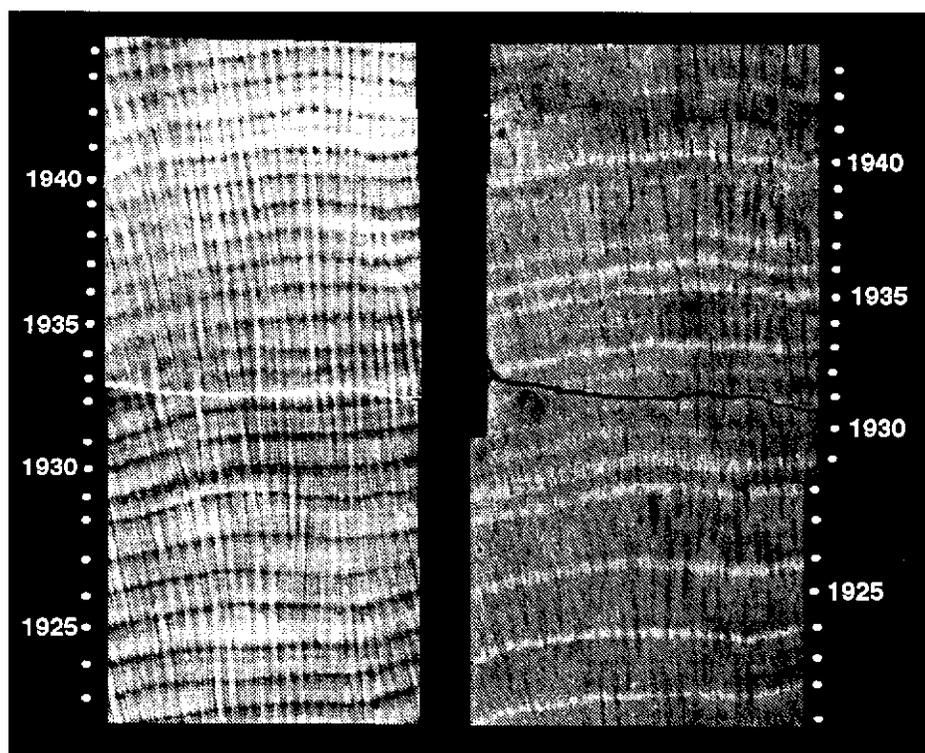
---

#### Reference

Gladfelter, E.H., Monahan, R.K., Gladfelter, W.B. (1978) "Growth rates of five reef-building corals in the northeastern Caribbean," *Bulletin of Marine Science* 28:728-734.

*Core samples:* A historical record of coral growth can be obtained from cores of head corals that have skeletons with annual density bands, similar to tree rings. These species include *Montastraea annularis*, *M. cavernosa*, *Diploria strigosa*, *Siderastrea siderea*, and *Solenastrea bournoni*.

The core sample is cross-sectioned parallel to the growth axis with a rock saw to produce slabs about 4 mm thick. X-radiographs of the slabs will reveal density bands and allow estimates of annual growth. The slabs can also be analyzed for a variety of chemical constituents that may provide information about the history of contaminants and other sea water characteristics. For example, freshwater may contain fluorescent soil acids that are transported to coastal reef areas during periods of high runoff and incorporated into the growing skeleton. These fluorescent bands will show up when the slab is illuminated with UV light.



*Annual growth bands in Montastrea annularis*

Coring requires use of an underwater hydraulic drill, pumps and coring equipment and is therefore a costly monitoring method. You may be able to get a hospital laboratory to produce the X-ray. Holes left by coring should be filled with Portland cement.

#### Reference

Halley, R.B., Curry, R. (1993) "Coastal Geology and National Parks: An example from Biscayne National Park," *Park Science* 13(1):4-5.

## COMPARISON OF MONITORING METHODS

Quadrats, photoquadrats and chain transects provide alternative ways to obtain and document the information needed to measure percent cover, species diversity, and relative abundance. As summarized below, each method has its advantages and limitations. Ideally, a coral reef monitoring program will include more than one method where appropriate. For more about using photoquadrats, see "Underwater Photography," III-29. Chain transects are described in more detail in the next section.

	Quadrats	Photo-Quadrats	Chain Transects
Equipment	Relatively inexpensive	May be very expensive, depending on equipment used	Relatively inexpensive
Difficulty	Relatively simple, but at least for initial survey must be done by someone who can identify species in the field	May be difficult to set up depending on equipment used, but simplest methods can be done by non-specialists	Tedious and exacting; must be done by specially trained divers
Damage to reef	Slight risk in areas of high relief, especially if grid is used	Depending on equipment used, may be risky in topographically complex areas	Even well-trained divers find it difficult to avoid causing some damage, especially in areas with branching corals
Data obtained	If grid is used, can provide reasonably accurate measures of percent cover, species diversity, relative abundance, density and size	Can be used to estimate percent cover, species diversity, relative abundance, density and size	Measures all surface areas below line to determine percent cover, species diversity and relative abundance; estimates spatial index
Limitations	Cannot be used to measure spatial relief; provides data only on projected surface area; difficult in elkhorn or staghorn-dominated areas	Cannot be used to measure spatial relief; provides data only on projected surface area; unsuited to areas with large or abundant octocorals that conceal other species	Cannot be used to directly measure species density or colony size; not suited to areas where stony corals are widely-spaced and small; impossible in elkhorn or staghorn-dominated areas
Use of data	Data are ready to use when diver leaves the water	Measurements cannot be determined until after photographs have been digitized	Data are ready to use when diver leaves the water
Replication of survey	Relatively easy, if done by the same person each time or by people who have been trained together	In permanent photoquadrats, precision depends on apparatus used and ability to take photo from exactly same spot	Even with well-marked transect, impossible to position the chain exactly the same each time
Calculating percent cover	Can be easily calculated, manually if necessary	Digitizing is time-consuming to do manually and difficult without access to computer and software; use of random dots also time-consuming	Can be easily calculated, manually if necessary

### Reference

Ohlhorst, S.L., Liddell, W.D., Taylor, R.J., Taylor, J.M. (1988) "Evaluation of reef census techniques," Proceedings of the 6th International Coral Reef Symposium 2:319-324.

## QUADRATS

The term “quadrat” generally refers to a square or rectangular sampling unit within which organisms are counted or measured, or to the frame which marks this area. Quadrats can be used to estimate percent cover of each species or other reef components and obtain information about density, abundance, diversity, and colony size.

**Limitations:** Quadrats are generally preferable to linear transects in monitoring Octocorallia and smaller stony corals. However, because a quadrat provides data only on the horizontal plane of the reef surface and not on spatial relief, its use is inherently problematic on an irregular and highly three-dimensional reef surface. Taxa with plate-shaped morphologies tend to be over-represented relative to columnar species such as “pillar coral” (*Dendrogyra cylindrus*) in census data, while cryptic taxa tend to be omitted completely. In these situations, linear transects may be more appropriate.

### Installation

**Size:** Although one-meter square quadrats are frequently used, the size of the quadrat may depend on the size of the organisms you are monitoring. While phycologists often use  $\frac{1}{4}\text{m}^2$  frames, coral biologists often use  $\frac{1}{2}\text{m}^2$ ,  $1\text{m}^2$ , or larger.

**Locution:** The quadrats should be far enough apart so that their boundaries do not overlap. The problem of parallax, in which the size and location of an object is affected by the angle from which it is viewed, may be exacerbated if a series of quadrats abut each other. Quadrats can be permanently or randomly placed to obtain general data on reef conditions. Quadrats can be sampled along transects by placing the frame on alternate sides of a line or centering it along the line.

To mark a permanent quadrat, you can use concrete nails or re-bar stakes at two corners on the diagonal or at all four corners of the quadrat. If you are using a quadrat frame or grid, the pins should rest on the inside of the corners. For more information about marking the site, see “Site Selection”, I-15.

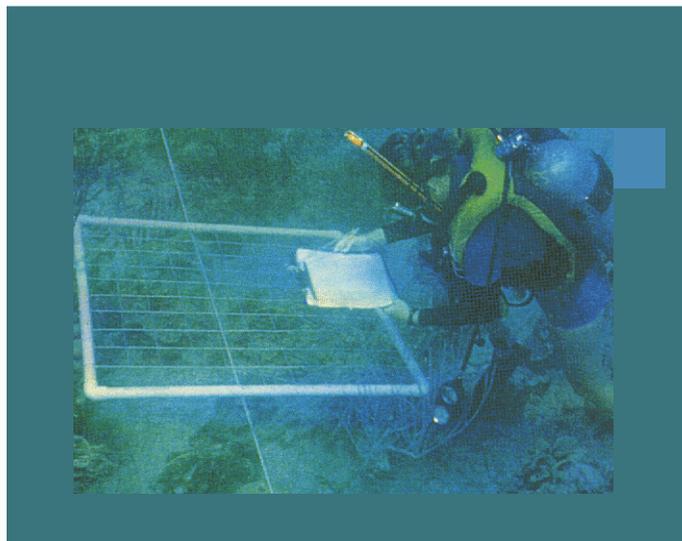
**Construction:** Small quadrat frames can be made from iron re-bar or stainless steel. For larger quadrats, in which weight becomes more of an issue, aluminum or PVC pipes are easy to handle underwater and less likely to damage the substrate. To decrease buoyancy and reduce resistance in carrying the frame through the water, you can drill small holes in it. The frame is usually removed from the site between monitoring visits.

**Quadrat grids:** To simplify measurements on relatively flat substrate, you can create a grid on the quadrat with string. For example, a 1-m quadrat can be strung with 10 vertical and horizontal lines to create a grid of 100 squares, with each 10-cm square representing 1% of the quadrat. By counting the number of squares (or fractions of squares) occupied by each species, you can estimate abundance and percent cover within the quadrat. In areas where the topographical complexity makes using a quadrat grid difficult, you can create a reference scale by drilling a hole or marking off every 10 cm on each side of the frame with paint.

## Data Collection and Analysis

**Percent cover:** For quadrats with relatively few species and little live coral and plant cover, you can estimate cover for each species while in the field by counting the number of squares (or partial squares) covered by each species and recording these numbers as shown on the sample data sheet below.

Sample Data Sheet	
Quadrat #5	# of Squares
A. agaricites	1.0
P. astreoides	0.7
S. siderea	4.0
M. annularis	2.0
P. astreoides	6.5
A. agaricites	5.0
D. strigosa	10.0
S. siderea	8.8
macroalgae	20.0
sand	42.0



*Using a Quadrat Grid*

If the quadrats contain several species and more live cover: to reduce the likelihood of error, you can record the data square by square on a grid written on your underwater slate, and tally the squares after the dive.

From the data in the above example, you can calculate the number of coral species (5), and the total live coral cover (38%) by adding up the number of squares occupied by each coral species ( $1 + 0.7 + 4.0 + 2.0 + 6.5 + 5.0 + 10 + 8.8 = 38$ ; space occupied by algae and sand are not calculated for live coral cover). However, more typically you would use data from several quadrats and derive average percent cover, etc. (Before doing parametric statistical analysis (which assumes a bell-shaped distribution), percentage data from quadrats and transects must be arcsin transformed. For more information about statistical analysis, see Appendix B.)

**Frequency and density:** You can calculate the “frequency” of a species by counting the number of quadrats in which the species is observed and dividing by the total number of quadrats. The “density” is the number of different species or colonies found within a given area, usually per square meter. It is important to remember that the number of coral colonies can be independent of species diversity and coral cover. For example, a hurricane that causes significant mortality (**loss** of cover) may bring about an increase in the number of coral colonies because of fragmentation, but the colonies would be of a smaller size. Or, two reefs may have the same number of colonies, but entirely different percent cover.

**Species diversity:** Increasing concern about the loss of species and degradation of ecosystems has led to an interest in measuring genetic, species, and ecosystem diversity, known collectively as “biodiversity”. In coral reef monitoring, most of the data collected pertains to the number of species, sometimes called “species richness”, and their relative abundances. The Shannon-Weaver index ( $H'$ ), which combines both number of species and relative abundance, is calculated using this formula:

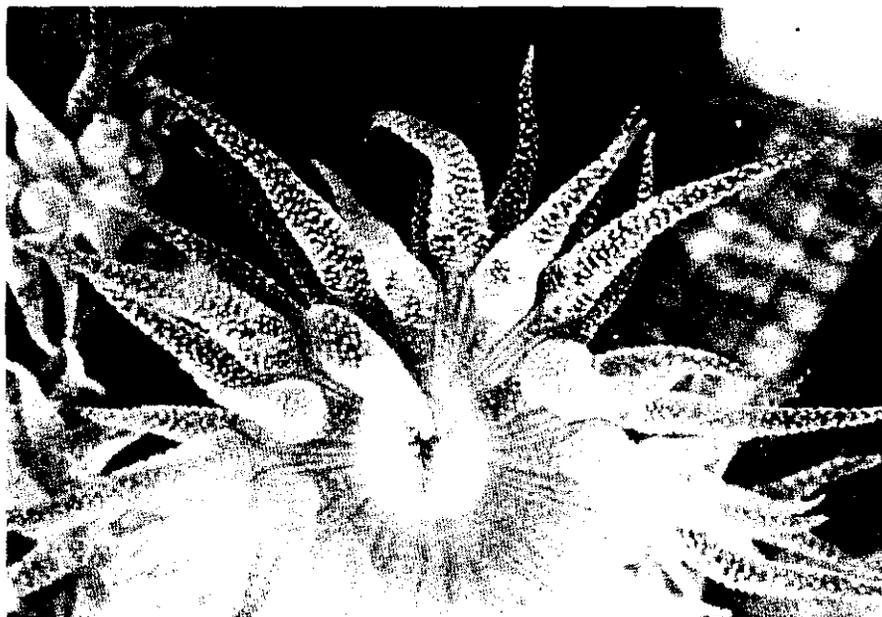
$$H' = -\sum p_i \ln p_i$$

where  $p_i = n_i/N$ , in which  $N$  = the total number of colonies of all coral species in the quadrat (or total number of centimeters of live coral under the line, for a chain transect), and  $n_i$  = the number of colonies of each coral species in the quadrat (or the number of centimeters for each species).

#### Reference

Shannon, C.E., Weaver, W. (1949) *The Mathematical Theory of Communication*, University of Illinois Press, Urbana.

**Quadrat grids vs. photo-quadrats:** You can get the same information about projected area from a quadrat on which a grid has been strung as from the two-dimensional view provided by a photograph (see “Underwater Photography,” III-29). However, certain calculations may take longer if done underwater rather than from a photograph, and use of a quadrat grid requires expertise in species identification for each sampling. With photographs, species identification may only need to be done on-site for the initial survey, so subsequent photos can be taken by any competent diver. Photography can also be combined with the use of a grid quadrat.



## CHAIN TRANSECTS

A linear transect is a line of a specified length laid out within a study site. Transects are generally positioned parallel to the shore, along depth contours (e.g., at 5, 10, and 15 meters). A transect laid perpendicular to shore may be appropriate if you want to include different reef zones (or depths) in the same transect. Preliminary transects can be used to delineate the different zones within your study area, or to determine the necessary length of permanent transects for long-term monitoring. Measurements can be taken along the entire surface beneath the line, using a chain as described below, or at specified intervals, as explained under "Line and Point Intercept Transects," III-21.

A chain transect is a relatively inexpensive and accurate way to gather information on species diversity, the relative abundance of different species, and the amount of hard substrate or sand. It is most effective for documenting changes in abundant larger coral species, and best suited to areas dominated by head (rather than branching) corals. Information on the percent cover by all major reef components can be calculated easily on a computer, or by hand if necessary.

By following the surface contour of the reef, chain transects provide data that you can use to estimate the "spatial index" of the reef: the ratio of reef surface contour distance to linear distance. As part of a long-term monitoring program, the spatial index provides a way to quantify changes in the topographical complexity of the reef.

### Limitations

Although chain transects provide certain information not available from quadrats, they do have certain disadvantages.

- Chain transects are inappropriate in *Acropora palmata* (elkhorn) zones, octocoral-dominated pavement areas, or in areas where stony coral colonies are widely-spaced and small. However, if the sampling chain crosses over an octocoral holdfast, it should be included in the data. The amount of living coral in dense *A. palmata* zones is probably best estimated with photographs.
- The chain transect method, also known as the "ball and chain method", is tedious and time-consuming. It's not unusual to spend over an hour on 10 m of transect.
- The reef may be damaged if the chain becomes entangled in branching coral.
- It's impossible to position the chain in exactly the same location each time. Changes noted during repeat sampling may reflect shifting in the chain's position rather than actual changes in cover on the reef; however, these shifts are unlikely to cause significant changes in results for the most abundant organisms.

## Data Collection

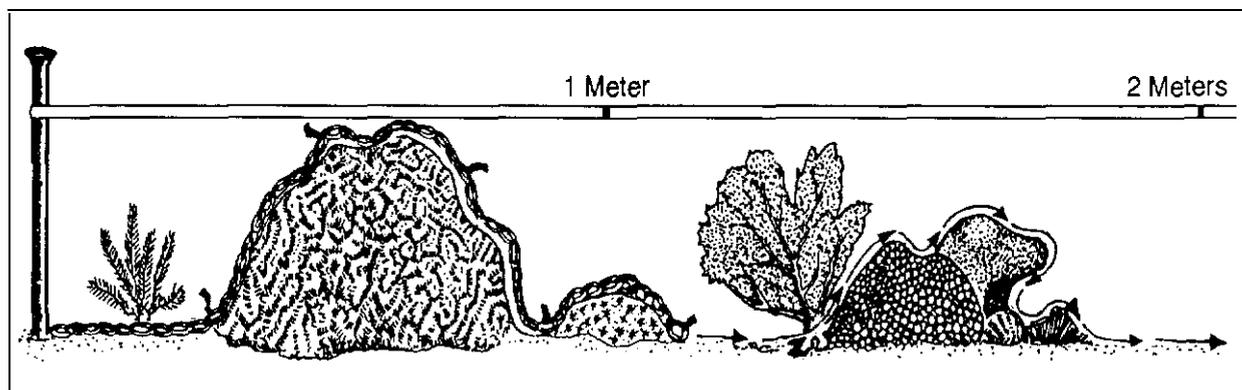
- 1) **Mark the transect.** For a preliminary transect, tie each end of a line marked off in meters or a waterproof fiberglass tape measure (20 to 30 m is usually sufficient) to a piece of dead coral along the depth contour at a randomly selected site. Keep the line as taut as possible and at the height of the tallest feature along the transect (usually a coral colony), and try to keep to the selected depth as much as possible (within a few feet). If the line is too high above the substrate, parallax will make your work more difficult and your data less accurate.

Note: Three-strand nylon lines stretch when wet, so check the measurement after the line has been soaked. To avoid stretching, use a polypropylene or double-braid nylon line, or fiberglass tape measure.

Permanent transects should be marked with metal stakes to indicate the exact beginning, middle and end of each transect. Attaching bright-colored flagging tape to the stakes will make them easier to find for repeat sampling. To help position the line in the same place each time, it's also a good idea to mark off every five meters with a nail, or other small reference marker. (See "Marking the Site," I-16.)

- 2) **Position the chain.** Drape a light-weight chain (such as a dog chain) over, around, and under all natural fixed surfaces directly below the line. A 5-foot length of chain with 1.3-cm links (77 per meter) works best in many situations. Smaller links take longer to count, and larger links may miss certain features. Be sure to use chains with the same size link each time you sample. Sampling progresses in 1-m increments, recording at which meter the data are collected. Periodically check your position by holding the chain against the tape and then letting it drop to the substrate.

Suggestion: You can prevent errors in counting links by marking off every 10 links with small pieces of brightly colored flagging or surveying tape.



*Positioning the Chain*

- 3) **Measure the surfaces.** Count the number of links it takes to outline the surface of each species or reef component that is of concern to you. If your objective is to determine the appropriate length for permanent transects and you are primarily interested in stony corals, you need to record only cover by live coral (by species); all other surfaces, living and dead, can be put into the category of “other” or “non-coral”. Include the fire coral *Millepora* (also to species).

For permanent transects, you may want to include reef components such as sponges, bleached coral, dead coral with algae, rubble (pieces of branching corals or plates, rather than intact dead coral heads), macroscopic algae, octocorals (holdfasts only), sand, and pavement (hard carbonate substrate of low relief, sometimes dominated by octocorals).

The chain must touch a solid structure at all times, even if it must be held up against the underside of a coral branch or ledge. Do not attempt to measure mobile organisms with links! But you may want to make note of important herbivores such as the spiny sea urchin *Diadema antillarum* when they are under or near the line. The substrate below the urchin is what should be counted in the transect data.

The objective: to measure as carefully as possible the surfaces under the line, even when there are several layers. For example, if an *Agaricia* is positioned over a *Montastraea* colony and directly under the line, you measure the live upper surface of the *Agaricia* plate, the dead undersurface, and the top of the *Montastraea* colony.

- 4) **Record the data.** Using mylar sheets or waterproof paper on a clipboard or an underwater slate, record the number of links for each category in each meter of line (not each meter of chain). For each meter of line, check to make sure that you’ve recorded at least the number of links equivalent to 1 meter.

Sample Field Notes			
Date: _____			
Location: _____		Depth: _____	
Divers: _____			
Surfaces -- # Links			
Meter 1:		Meter 2:	
Ma	5	Aa	-- 5
sand	-- 25	Ds	-- 7
Aa	-- a	maca	-- 4
dca	-- 24		
rubble	-- 35		

- ▶ Record each colony or other surface as you come to it along the transect; don't combine separate colonies of a species and record a total for the species within each meter. Abbreviate species names, being careful not to use confusing abbreviations, e.g., *M.a.* could be ***Montastraca*** annularis or *Millepora alcornis*.
- ▶ If hard substrate or dead coral is covered with sediment less than 1 cm thick, refer to this portion of the transect as "pavement" or "dead coral with algae".
- ▶ If sediment on pavement or dead coral is at least 1 cm thick, record it as "sand".
- ▶ "Macroscopic algae" (abbreviated "maca") refers to conspicuous algal plants such as the brown alga *Dictyota* and red alga *Martensia*, whose fronds project more than 1 cm above the substrate. If possible, identify the alga to genus or species.
- ▶ Dead corals with turf algae (abbreviated "dca") often make up a larger portion of the transect than live corals.

If time and energy permit, you can check the variability among different observers or the same observer's results when repeating a transect by doing each transect two or three times. It is usually adequate to survey the transects every six months.

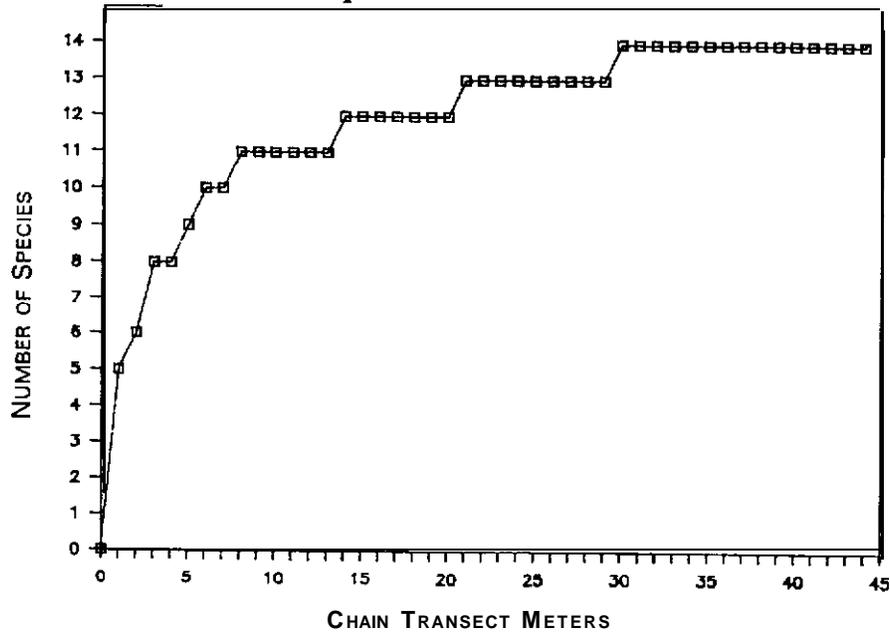
This process requires a major investment of time, but gives you a lot of information in return. Chain transects are much more difficult in some areas than others. Don't give up! If you encounter small caves or big holes, try to position the chain so that you can estimate the amount of dead coral or sponges, etc., in these places. If you come upon a deep sand channel more than two meters across, make note of it and start recording where the reef itself begins again.

## Data Analysis

**Preliminary transects:** To determine the appropriate length for a permanent transect, plot the cumulative number of coral species (or other species of interest) against the number of meters along the preliminary transect line. After a certain number of meters, depending on the site, there will typically be a leveling off as the number of additional species decreases. At sites around St. John, 20-m transects have been appropriate, while scientists in Florida have found that 25-m transects were preferable.

After analyzing the results from the preliminary transects, you can establish permanent transects placed randomly along each of the selected depth contours within selected study areas or zones.

### *Species Area Curve*



#### *Percent cover and structural complexity:*

- To determine the percent of live coral cover or other component, divide the number of links for that component by the total number of links in the transect.

$$\text{Percent live coral cover} = \frac{\text{No. of links live coral}}{\text{Total no. of links along transect}} \times 100$$

- To determine an index of topographical complexity calculate the ratio of the length of the chain (in centimeters) to the length of the line.

$$\text{Structural complexity} = \frac{\text{No. of transect links} \times \text{cm per link}}{\text{Total transect length (cm)}}$$

The preceding calculations can be done with a calculator, but they are easier if you have a software package such as Lotus or Microsoft Excel. Biologists in Virgin Islands National Park have developed a simple Lotus-based program to analyze chain transect data and will provide a copy on request. A more sophisticated software program that can also calculate the species diversity index  $H'$  is available from Dr. James Porter, University of Georgia. For information about statistical analysis, see Appendix B.

#### Reference

Rogers, C.S., Gilnsck, M., Fitz, H.C. (1983) "Monitoring of coral reefs with linear transects: a study of storm damage," *Journal of Experimental Marine Biology and Ecology* 66:285-300.

## LINE AND POINT INTERCEPT TRANSECTS

Line intercept and point intercept transects are other ways to gather data that can be used to estimate percent cover, relative abundance, and diversity. The intercept method is simpler and quicker than using a quadrat or a chain transect, so you can survey a larger area in the same period of time. However, like the quadrat, intercept transects cannot be used to calculate a spatial index, and like the chain transect they may not provide accurate sampling in areas where coral colonies are small and widely scattered.

To do a line intercept transect, secure a fiberglass tape measure to both ends of the transect with the tape draped over the reef in between, and record each species or substrate component and its length under the tape. In a point intercept, you record only what is located at each specified point along the line, e.g., every 20 cm. Depending on your objectives and available resources, you may want to survey several lines at each site, use a longer line, or longer intervals. To test the adequacy of your sample size, check the cumulative species recorded against the number of transects surveyed. The number of points scored by a given species divided by the total number of points yields the percentage cover.

### Reference

Ohlhorst, S.L., Liddell, W.D., Taylor, R.J., Taylor, J.M. (1988) "Evaluation of reef census techniques," Proceedings of the 6th International Coral Reef Symposium 2:319-324.

## OCTOCORALS AND SPONGES

Octocorals (soft corals) and sponges are important components of the reef. Octocorals, a group of Cnidarians (Coelenterata) that includes "gorgonians", occur in most reef habitats, and dominate some reef zones. Their abundance often varies inversely to stony coral cover. Species identification can be tedious and difficult, but it's worth making the effort to learn to identify them in the field so you can avoid destructive sampling. Valuable information can be obtained from surveying octocorals at the level of families or genera rather than species.

**Quadrats:** To survey octocorals, quadrats of one square meter are standard, but 0.25-m<sup>2</sup> quadrats are also used. Still photography can be used to identify specimens, growth of new recruits, and progression of disease. Photo-quadrats and grid quadrats are inappropriate for most habitats because octocorals occur in widely ranging sizes and tend to overlap, preventing proper placement of a grid. In addition, large octocorals may conceal other benthic organisms in a photograph.

*Belt Transects:* For a quick assessment of octocoral abundance and distribution, you can look at the species within a belt transect of specified width across the study area, e.g., 1 meter wide and 20 to 25 meters long. Along depth contours, use fiberglass tapes with the ends secured. Count and identify (to lowest possible taxa) all octocorals within each linear meter of transect. Twenty transects are generally sufficient for adequate sampling in the Caribbean.

*Data to Record:* Species identifications, number of colonies, height measurements, and number of newly settled recruits ( $\geq 5$  cm tall).

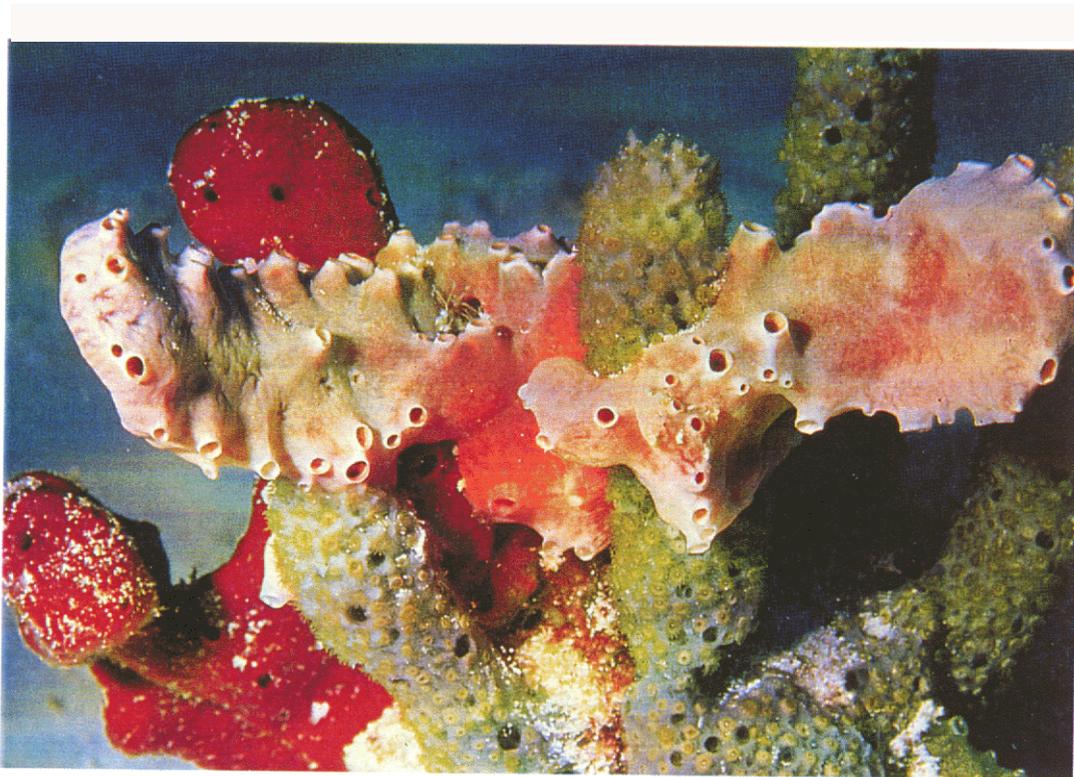
#### References

- Kinzie, R.A. (1973) "The zonation of West Indian gorgonians," *Bulletin of Marine Science* 23(1):93-155.
- Goldberg, W.M. (1973) "The ecology of the coral-octocoral communities off the southeast Florida coast; geomorphology, species composition, and zonation," *Bulletin of Marine Science* 23(3):465-488.



Sponges: While there are about 40 species of corals in West Indian reefs, there are about 300 common species of sponges. They are critical constituents of the reef. For example, they help construct the reef framework (sclerosponges); they contribute significantly in creating reef sediments through bioerosion and spicule formation; and their varied forms of growth and abundance increase topographic complexity, which enhances local diversity. In addition to being a net primary producer of oxygen, a single species such as the chicken liver sponge may contribute 50-120% of the nitrogen required to sustain reef productivity. This species is frequently eaten by certain reef fish (mostly angel fish), and it is the most important food item for the endangered hawksbill turtle.

Sponges can generally be sampled along with other attached benthos in quadrats or chain transects (see previous sections). Although some species are identifiable in the field, many can only be identified in the lab.



#### Reference

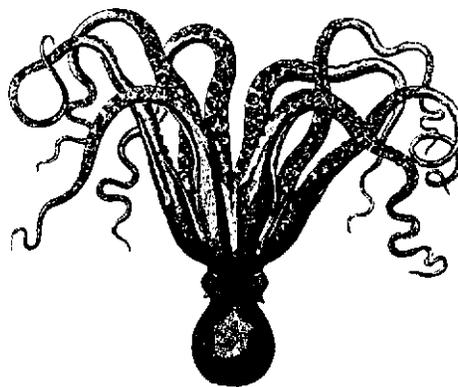
Vicente, V.P. (1982) Common sponges. In: Kaplan, E.H. *A Field Guide to Coral Reefs (Caribbean and Florida)*, The Peterson Field **Guide** Series, Houghton Mifflin Company, Boston. pp. 127-133.

## ALGAE

Many reefs in the Caribbean and western Atlantic have relatively low amounts of live coral cover (often less than 40%) and high amounts of cover by algal turf and macroscopic algae. But the biomass of macroscopic algae can vary widely across a reef and over time, often seasonally. Macroalgae populations may fluctuate depending upon local nutrient fluxes (sometimes associated with heavy rains or upwelling) and temperature changes. However, dramatic increases in algal cover and biomass may indicate an increase in nutrients from sewage or agricultural fertilizers, or a decrease in grazing by herbivorous fish or sea urchins.

Algae can be loosely grouped into the four categories shown below. The “1 cm” distinction between macroscopic and turf algae is somewhat arbitrary.

Macroscopic algae (macroalgae)	“Fleshy” algae which are not hard to the touch and project more than 1 cm above the substrate.
Calcareous algae	A wide range of red and green algal species that are hard to the touch; <i>Halimeda</i> is often the most common genus.
Crustose coralline algae	Appear as hard smooth pavement covering small or large area; color varies from dark pink to purple, or may show a grayish hue; do not confuse with hard bare substrate that tends to be yellowish or whitish in appearance.
Turf algae (algal turf)	May look fleshy and/or filamentous but do not rise more than 1 cm above the substrate; <i>Gelidium pusillum</i> and <i>Coelothrix irregularis</i> are common species.



## Algal Biomass

**You** can make a rough estimate of algal biomass (wet) expressed as g/m<sup>2</sup> by removing all of the algae within a 0.25-m<sup>2</sup> quadrat. To obtain an adequate random sample, you'll need to collect and weigh the algae from at least 15 quadrats.

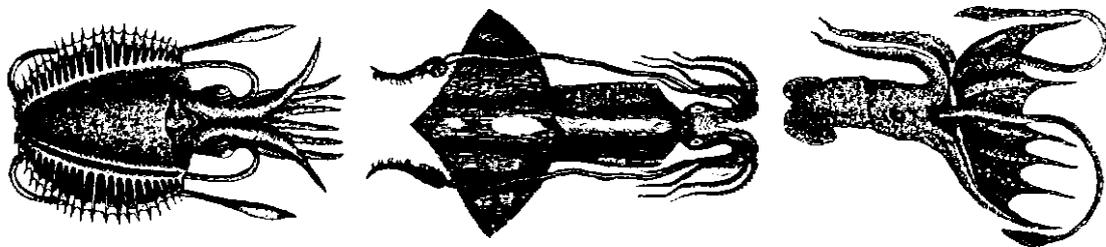
To estimate algal biomass (dry) of endolithic forms (algae growing in the substrate itself), you can use settling plates made of natural substrate (e.g., cross-sections of elkhorn coral), or a variety of other substances such as ceramic tiles.

---

### Estimating Algal Biomass

- 1) Secure at least three settling plates to dead coral, or attach them to a survey stake, re-bar or other apparatus and leave them at the site. Algae will usually begin to grow on the plates within a week. (See method for monitoring recruitment of benthic organisms in the next section.)
- 2) Use a grid and three pairs of random numbers to select three 1-cm<sup>2</sup> subsamples on each plate. Scrape the subsamples to a depth of 1 mm and preserve them in a 1% formalin solution, combining all subsamples from the same plate. Decalcify the sample using a dilute acid (5% HCl or HNO<sub>3</sub>).
- 3) Filter the sample onto pre-weighed filters, rinse it with deionized water, and dry it to a constant mass at 60°C.
- 4) To determine the g/cm<sup>2</sup> of biomass, divide the difference between the final filter weight and the initial filter weight before filtration by the number of 1-cm<sup>2</sup> subsamples that were filtered (in this example, 3).

$$\text{g/cm}^2 \text{ biomass} = \frac{\text{Final weight} - \text{initial weight}}{\text{No. of cm}^2 \text{ of subsamples}}$$



## Algal Species Composition

In determining algal species composition, you want to be sure to get an adequate sampling of macroscopic and algal turf communities. Here are some sampling methods.

...

### Estimating Turf Algae Species Composition

- 1) As described for estimating biomass, scrape four 1-cm\* subsamples from each plate, preserve the sample in 1% formalin solution, and decalcify it.
- 2) Mount all subsamples from the same plate on the same slide, and note the algal species present while scanning the slide at 100x magnification.
- 3) To estimate relative abundance, put a 10x10 ocular grid over the slide and count the intersections on each species in at least 50 viewing fields.
- 4) To calculate the percent relative abundance for a species, divide the total intersections for that species in all viewing fields by the total intersections for all species in all viewing fields and multiply by 100.

$$\%relativeabundance = \frac{\text{No. of intersections for a species}}{\text{Total intersections for all species}} \times 100$$

**Macroalgae:** To estimate the macroalgal species composition on a setting plate, count the intersections of an 8cm x 8cm grid (64 total points) located on top of each macroalgal species.

**Photo-quadmts:** You can estimate algal abundances using a Nikonos camera with a 28-mm lens and close-up frame. Project the resulting 35-mm color slides onto an 8½x11 sheet of paper with randomly located dots. (See "Dot Grid," III-34.) The number of dots falling on each algal component — algal turf, crustose algae (primarily corallines), calcareous algae and macroalgae — can be summed and expressed as a percent of the total algal cover.

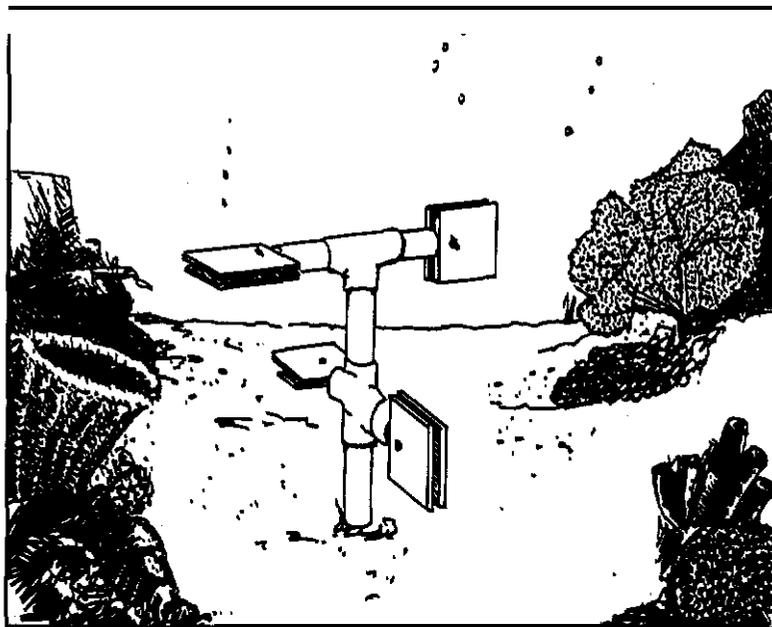
#### References

Bohnsack, J.A. (1979) "Photographic quantitative sampling of hard-bottom benthic communities," *Bulletin of Marine Science*, 29(2): 242-252.

Carpenter, R.C. (1990) "Mass mortality of *Diadema antillarum*; I. Long-term effects on sea urchin population dynamics and coral reef algal communities," *Marine Biology*, 104:67-77.

## RECRUITMENT OF BENTHIC ORGANISMS

Recruitment is the influx of new members into a population by reproduction or immigration. The establishment of new coral recruits generally indicates good conditions for reef development and growth. For overall reef assessment, photographs taken every three months are usually adequate to document recruits more than 5 mm across. You can also examine recruitment on settling plates that are secured to the substrate or tied to a sampling apparatus in a "tree" with four horizontal branches. The following instructions explain how to construct the kind of permanent recruitment sampling apparatus shown below.



*Recruitment Sampling Apparatus*

## Constructing a Recruitment Sampling Apparatus

- 1) Using PVC cement and pipe arrays (Schedule 40, 1 1/2-inch diameter) designed to fit over stainless steel reference stakes, glue two 17.8-cm (7-inch) pieces of pipe into opposite ends of a cross fitting. Insert a T fitting into the top pieces.
- 2) Glue a 9.9 cm (3.5 inch) piece of pipe with a 10.8 x 3.8 x 0.64 cm (4 1/4 x 1 1/2 x 1/4-inch) length of PVC flat stock into the open ends of the T and cross fittings.
- 3) For each recruitment array, bolt four pairs of 10.8 cm square ceramic tiles (4 1/2 inch) to the flat stock piece with 1/4-inch stainless steel hardware so the unglazed surfaces are exposed.
- 4) Arrange two pairs horizontally and vertically on an array to help determine whether organisms prefer particular orientations for settlement.
- 5) Secure the array to the stainless steel stake using two 1/4-inch set screws.
- 6) Every 6 or 12 months (depending on your study objectives) remove a pair of tiles for examination and replace them with clean tiles.

After collecting the settling plates, examine them using a dissecting scope. Stony corals and other organisms on the plates should be identified to the lowest possible taxon and enumerated to determine abundances and densities, e.g., the number of recruits of each species per settling plate, or per square meter.

### References

Rogers, C.S., Fitz, H.C., Gilnack, M., Beets, J. Hardin, J. (1984) "Scleractinian coral recruitment patterns at Salt River Submarine Canyon" (St. Croix, U.S. Virgin Islands), *Coral Reefs* 3:69-76.

Jaap, W.C., Wheaton, J.L., Donnelly, K.B., Kojis, B.L., McKenna, Jr., J.E. (1992) "A three-year evaluation of community dynamics of coral reefs at Fort Jefferson National Monument, Dry Tortugas, Florida, USA," Proceedings of the 7th International Coral Reef Symposium.

## UNDERWATER PHOTOGRAPHY

### Uses

Photography is useful in documenting general reef conditions, changes in reef structure, and the effects of natural and human-caused damage. The variety of sampling techniques available makes it possible to use photography for either a quick qualitative assessment of reef changes or a detailed quantitative analysis. Videotapes are especially useful for showing park managers and non-divers the marine environment.

Photographs and videotapes provide a record of community composition and spatial arrangement of reef organisms that is not available from other forms of data, and that may become of great importance later. For example, although no one anticipated the 1983 die-off of the black sea urchin *Diadema antillarum*, photographs indicate its previous density. In many cases, a photograph will speak more loudly and clearly than a statistical report on changes in percent cover over time. A picture can be worth a thousand numbers.

As a monitoring technique, photography also has the advantage of being relatively easy for volunteers or others who are not trained reef biologists. For example, dive tour operators visit the same dive spots frequently and can provide useful observations on reef conditions and photograph specific sites every few months to document change.

### Limitations

Although photography can provide much valuable information, it has limitations that need to be considered when you design a monitoring program and choose your sampling sites.

- Organisms under coral plates or rock ledges will not be visible in a photo, and even those organisms which are visible cannot always be readily identified in a photograph. On-site observation is needed to distinguish between certain species.
- Obtaining quantitative information from photographs of areas with large or abundant octocorals is difficult because they overshadow other organisms.
- Because photographs provide only a two-dimensional view of the reef, they cannot be used to estimate spatial relief. However, stereophotography will provide three-dimensional photographs that can yield information on relief. It is technically more complex and requires sophisticated analytical systems.
- To accurately detect small changes within a small area, you must photograph the area from exactly the same spot each time. Shifts in coral heads or rubble due to storms or bioerosion can make this almost impossible. (This problem can be minimized if a monopod is used, as explained later.)
- Corals may be damaged if you place a quadropod over or near them, especially in topographically complex areas.

- Analysis of photographs to assess changes in percent cover by different species can be done through digitizing of the photographs (“image processing”). Manual digitizing is very time consuming, and many people do not have immediate access to the hardware and software necessary for computer digitizing. However, you can store the photographs (preferably in a cool dry place to avoid fungus damage) until you have the capability to analyze them yourself or send them to a colleague for analysis. (Although less accurate, you can also use a grid of random dots to estimate cover.)
- Photo coverage of large areas is problematic. If a photo is taken from a long distance away, the resolution and water clarity may not be sufficient to identify organisms. An alternative is to take a series of overlapping photos and create a photo-mosaic. Under optimal conditions, it is possible to make a repeatable and accurate mosaic.
- Any movement of the camera, frame, and other apparatus components will reduce the quality of the photograph. Photos taken on rugged topography are very difficult to match and align. An alternative to photomosaics is a series of independent photographs taken from fixed references.

#### **Appropriate Sites**

**There** are some special considerations in site selection if you plan to use underwater photographs or videotapes, especially if they are your only or primary means of data collection.

- ▶ The substrate in the site must be able to accommodate whatever permanent stakes will be needed to anchor a camera mounting apparatus.
- ▶ If the surrounding area has extreme topography, the depth of field established by the mounting apparatus will place limits on the photographic coverage.
- ▶ Photography can be used in high relief areas if qualitative analysis is sufficient for your purposes.
- ▶ Dense gorgonian areas should be avoided. A large sea fan can obstruct an entire photograph.

For more guidance on choosing study sites, see “Site Selection,” I-15.

## Replication

Long-term monitoring requires a process of data acquisition that is repeatable. If your objective is to precisely measure change at specific locations over time, you must make sure that your photographic coverage is the same each time. For example, you can:

- ▶ Use a mounting apparatus so that the camera is always positioned at the same angle and distance from the substrate (or subject), and mark the exact sites for placement of the apparatus.
- ▶ Attach the apparatus to a reference marker, or move it between two reference markers, so the photographic coverage is always the same.
- ▶ Include a ruler or other scale, compass, identifying number, and the sampling date in the photo or video image.

To see whether you can repeat the photograph exactly, repeat a series of photos at the same location when you do the first sampling and check them for consistency.

**Parallax:** When you view the same object from different angles or distances, its position and size may appear to change, a phenomenon known as “parallax”. In photography, if your camera is not properly aligned for a series of photos, parallax may also result in an overlap of frames. The longer the distance between the camera and the surface you are photographing, the greater the likelihood of parallax.

**Color variations:** Factors affecting color, such as daily and seasonal changes in available light and variability in strobe output, must also be taken into consideration when photographs are compared. To help document subtle changes over time, you can include a color chart that has gradations of the color characteristic of the species in the photo. Use consistent manual exposures instead of automatic settings for more consistent colors.

## Equipment

If you don't have the time, money, or expertise for sophisticated underwater techniques, it's still important to use whatever underwater camera equipment is available to photograph your study site at the outset and periodically thereafter, so you have a visual record that can be used to document major changes in the reef.

**Nikonos cameras:** Most of the methods referred to in this manual specify use of a Nikonos camera, a compact, rugged, and generally dependable waterproof unit that comes with an automatic light metering system and flash exposure. However, unlike a single-lens reflex (SLR) camera, the Nikonos models III-V require you to compose the shot through a viewfinder, not directly through the lens, and you must focus by measuring or estimating the distance. This is not a problem if you use a close-up framer. The newest Nikonos is an SLR camera (see next page).

**Nikonos close-up kit:** The Nikonos close-up kit, which comes with several "mini-framer" camera attachments and a threaded tripod mount, provides a simple way to document coral recruitment and monitor other changes in small areas with low relief. Either a 28-mm or a 35-mm lens can provide resolution of about 3-5 mm with Kodachrome 64 film. Permanent photo-quadrats located on hard substrate can be marked for resampling by pounding masonry nails into three corners of the photoframe, leaving at least 2 cm of nail above the substrate. Keep in mind that masonry nails will not last for more than two or three years underwater.

**SLR camera:** If you want a photograph of exactly what you see through the viewfinder, you may prefer to use an SLR camera in a waterproof housing with a dome port, so you can focus precisely. Although the resulting photographs can be excellent, this equipment is generally bulkier and more fragile than Nikonos cameras, and more difficult to attach to a mounting frame. Some SLR cameras also have auto-focus and auto-bracketing, and can imprint the date and time on the negative or slide. The Nikonos SLR camera does not require a waterproof housing, and a variety of lenses (including zoom) are available.

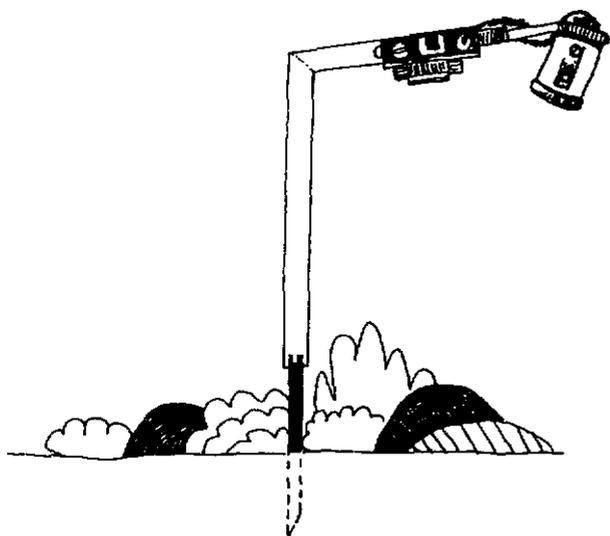
**Lenses and strobes:** Special lenses and strobe lights can help improve the quality of your underwater photos. Sea water acts as a filter and reduces contrast, especially diminishing colors with longer wavelengths; the farther the light travels through it, the more pronounced the effect becomes. Only blue and some green wavelengths extend below 250 meters. Underwater photography is therefore often done with a wide angle lens (15 to 28-mm) to get the camera as close to the subject as possible.

In clear shallow water, photographs can be taken using natural light, but as depth increases, an electronic flash unit (strobe) or other artificial light is necessary to provide sufficient light, accurate color and contrast. However, using a strobe to illuminate the subject will also illuminate suspended particulates, which may create white specks on the photograph. To minimize this effect, it's generally best to position the strobe so it concentrates light on the subject rather than illuminating the water between the lens and the subject. Use rechargeable nicad batteries to reduce costs. After 36 photos, the batteries should be fully discharged and then recharged.

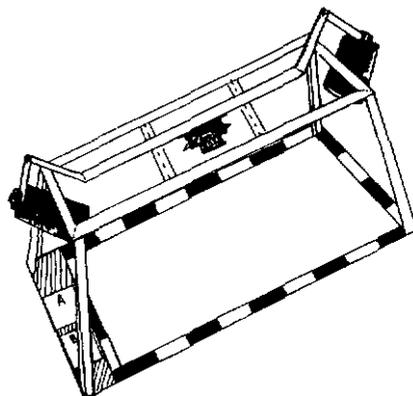
**Maintenance:** Underwater camera equipment requires meticulous care before and after dives to prevent corrosion. Use isopropyl alcohol to clean the lens, and rinse the camera (final wash) with distilled or deionized water to avoid salt deposits. Internal O-rings in the Nikonos camera and underwater housings should be replaced by the manufacturer or Nikonos repair facilities every 6-12 months.

**Film:** Your choice of film may depend largely on personal preference as well as on water visibility and the darkness of substrate. While more expensive, color film makes it easier to distinguish among components such as crustose coralline algae, encrusting foraminiferans, and bare rock, which may not be possible from black and white photographs. Higher-speed films (ASA/ISO of more than 100) are appropriate for natural-light photography that requires quick exposures (e.g., if there is unavoidable motion underwater), but they lack the fine grain of slower films. Having an artificial light source permits use of faster exposure speeds and slower film, which can result in sharper photos.

Mounting *apparatus*: To photograph larger areas for repeated sampling, you need an apparatus that will position the camera at a constant angle and distance, and a way to anchor the apparatus to the reef at each permanent site while you are taking the photos. Two of the most common methods use an aluminum or PVC *monopod* (anchored to one survey stake) or *quadrapod* (anchored either at opposite comers or at all four comers).



*Monopod*



*Quadrapod*

---

**Advantages** Less risky to reef, especially in areas where corals are large or abundant; easier to construct and faster to use.

**Limitations:** Since replication may be less precise, it is better for providing qualitative than quantitative data.

---



---

**Advantages** More stable; can provide better replication by minimizing the effect of surge or unsteady hands.

**Limitations:** Finding an appropriate site with substrate that can be marked (and drilled) at up to four corners may be difficult in some locations.

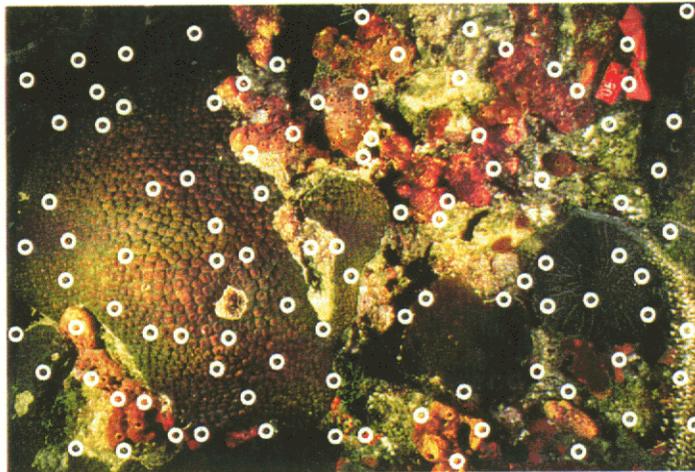
---

Although the dimensions of the apparatus should reflect the size of the colonies you are monitoring, your ability to maneuver a large apparatus underwater without injuring the reef is limited. Neither the quadrapod nor the monopod is well-suited to areas with very high relief. For suggestions on some specific mounting devices, see Appendix A.

## Analysis of Photographs

After the photos have been taken, various methods of differing sophistication are available to analyze them.

**Dot Grid:** If you place a grid of random dots over a slide image of a photo-quadrat on a back-lit projector, you can assume that the proportion of dots that lie on a substrate is equal to the proportional area of the substrate. Random dots are preferable because they do not require that each image be scaled. Each configuration of dots should be used only four times, but you can rotate the sheet to get four different configurations.



You can determine the optimal number of dots by conducting a preliminary statistical analysis. A grid of 100 dots is usually adequate for large organisms, but 200 is better statistically, and more dots will decrease the variance for some species. If a projector is not available, you can obtain similar results using a binocular microscope with a grid of random points photographed onto 35-mm film and placed beneath the slide.

**Digitizing:** A more accurate but time-consuming method of determining percent cover is to outline each coral colony or other organism in the photograph with a digital planimeter, or to use an electronic planimeter connected to a computer or image analyzer. Because a coral reef is three-dimensional, the outlined area is *projected*, not *actual* surface area. Changes in these areas over time indicate changes in total live coral cover and relative abundances of different species. The underside of colonies in view are not surveyed, nor are vertical surfaces.

Several software programs are available which can assist analysis by digitizing selected areas (such as coral colonies) within photographs. The program developed by Dr. James Porter and Ms. Linda Chiang of the University of Georgia uses Microsoft Quick Basic and a Jandel digitizer to analyze areas within photostations (see Appendix A, page 6). After tracing the outline of each coral colony onto mylar sheets from photographs, you digitize them to get estimates of projected surface areas. To obtain a copy of the manual containing a diskette with the software program, see "People to Contact," VI-7, for address and phone number.

Jandel's Sigma Scan is a popular software package that includes management of digitized data. Contact Jandel Scientific, 65 Koch Road, Corte Madera, CA 94925 for details.

Scanning: Another way to quantify photographic data is to scan the image into a computer data base. Scanning is less time consuming than digitizing; however, the hardware and software are relatively costly. Both slides and prints can be scanned but the images may have fuzzy boundaries, which can confound data processing.

### Reference

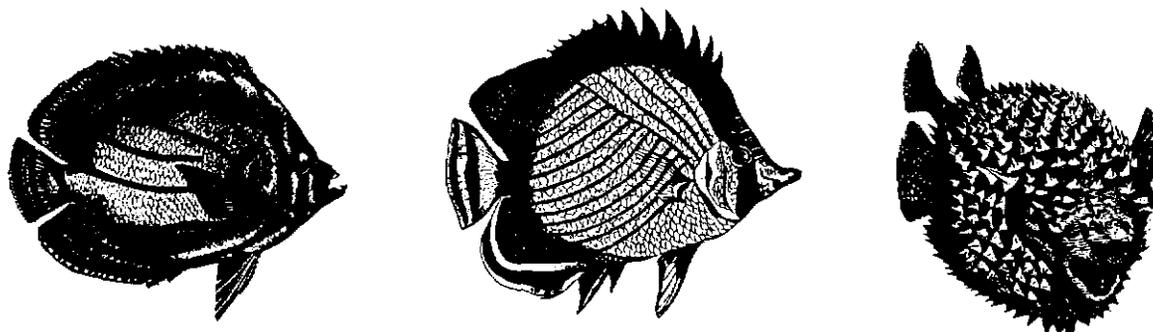
- Porter, J.P. (1990) **Methods for the Analysis of Coral Reef Community Structure;**  
1. Photostation, A. Field methods and data acquisition, Report to National Park Service.

## **Videotaping**

Videotaping has certain advantages over still photography for ecological monitoring. Although videotape is ideal if you want to photograph a large area quickly, it has limitations in comparative and quantitative analysis. Theoretically, two videos taken of the same area at different times can be run side by side, making a single video to observe gross changes. However, reproducing the exact path, speed, and distance from the substrate for repeated sampling of a video transect remains difficult. A "cable car" mechanism has been used to slide the camera over the substrate at a fixed distance to create a visual belt transect (see Appendix A, p. 7). Once the cable carriage apparatus is set up, it is relatively easy to use, but experience is required to get reliable results.

You should not underestimate the value of simply swimming around the reef with the video camera and recording. This method will provide qualitative information about the condition of the reef. Video images can be imported into graphic analytical programs with a frame-grabbing package that evaluates the various colored polygons. These tools, both the underwater video camera and computer analysis of video information, are expensive but can speed up data collection.

*Limitations:* Videotaping can generally provide only qualitative data. Because the distance between the camera lens and the reef surface is not constant, it's difficult to determine the relative scale of the components. To partially alleviate this problem, you can bolt a rod with a measurement scale to the bottom of the camera housing so that the image has a scale in it.



## CENSUS OF REEF FISHES

Reef fishes depend on reefs for food and shelter. In turn, reefs are affected by fish species that feed on macroalgae and algal turf (herbivores), and those that feed on coral polyps (corallivores). Through their waste, fish also provide an important source of nutrients, a very limited resource on coral reefs.

The main objectives for reef fish censuses are to compare fish populations among reefs and other habitats, and to quantitatively monitor species composition and relative abundance over time. For example, a reduction in the top predators (piscivores), declines in species abundance, and shifts to smaller average sizes may indicate fishing pressure. Fish censusing is difficult in coral reef environments because of the structural complexity of the habitat and the diversity, mobility, and abundance of reef fishes. Fish censusing also requires extensive training, as it may be necessary to recognize over 300 different species.

### Assessing Fish Populations

Different fish species that appear together are referred to as a "fish assemblage." Three aspects of reef fish assemblages that can be monitored are:

- . Diversity: the number of different species;
- ▶ Structure: species composition and relative abundance; and
- ▶ Population density: the number of fish of a given species per unit area.

Although attempts have been made to develop a single censusing method that will accurately measure all three characteristics, fish biologists generally agree that no such single method exists. You will need to decide what information is most important for your management needs and then select one or more appropriate methods.

### Census Methods

The most common methods for visual fish censuses are: stationary counts, belt transects, and random swim techniques. In choosing a method, be sure to consider the behavior of the relevant fish species (e.g., cryptic, schooling, attracted or repelled by divers).

- The **stationary census** (Bohnsack and Bannerot, 1986) focuses on the relative abundance and frequency of occurrence of all species observed at the site.
- The **belt transect** (Brock, 1954) method yields better density estimates and covers a larger area per census.
- The **random swim technique** (Jones and Thompson, 1978) provides more complete information on total species richness.

**Limitations:** All visual census methods have the following limitations:

- Observers generally underestimate the abundance of most species.
- Only the “observable” portion of the fauna is counted, so cryptic, nocturnal and pelagic species are most likely to be underestimated.
- Observers must be able to identify fauna quickly and correctly; expertise and consistency among observers is difficult to obtain.
- The presence of a diver will affect the behavior of fish.

**Frequency and Number of Censuses:** How many censuses, how many sites, and how often you should sample will depend on your monitoring objectives. Initially, you may want to census fish over several consecutive days to determine if there is any short-term variability. To detect seasonal changes in abundance and species richness, fish should be censused monthly until a baseline is established. To detect long-term changes sampling should be conducted at least once a year, at approximately the same time of day. It is preferable to census during the same month or at least in the same season each year, with at least 10 censuses conducted each time at each site. Statistical analysis may indicate more samples are necessary to optimize sampling. Methods for optimizing sample number appear in Green (1979) and Bros and Cowell (1987).

#### References

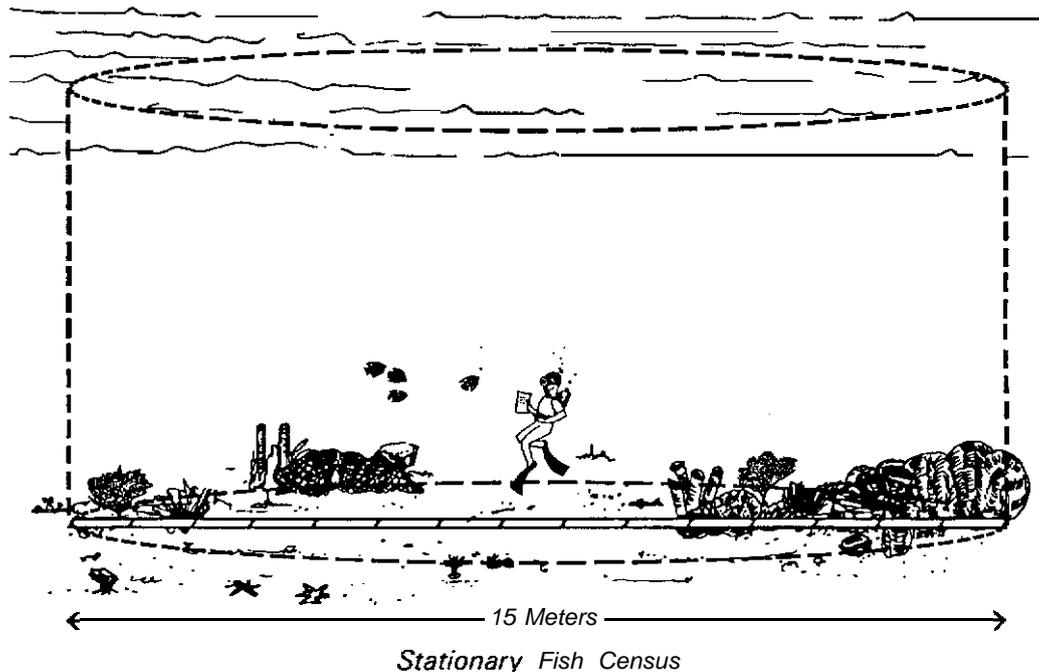
- Bros, W.E., Cowell, B.C. (1987) “A technique for optimizing sample size,” *Journal of Experimental Marine Biology and Ecology*, 114:63-71.
- Green, R.H. (1979) *Sampling Design and Statistical Methods for Environmental Biologists*, John Wiley & Sons, New York, 257 pages.

### **Stationary Fish Census**

The Bohnsack and Bannerot (1986) method has been very widely used throughout the Caribbean. The basic technique is presented here, although you may find a variation of this method better for your particular area. Appropriate modifications (e.g., changing the size of area sampled, the amount of time spent per census, how the time is allocated while underwater) will depend on local conditions (e.g., visibility, depth) and management needs.

- 1) **Establish a sampling radius.** At each randomly selected site, record the depth, maximum relief of the site, and percent cover by various bottom type classifications (e.g., sand, corals, algae, rubble, etc.). Stretch a tape measure out 15 meters along the reef to mark the sampling diameter; take up a position at the 7.5-m mark.

2) **Make a species list.** While rotating at the 7.5-m mark, scan the field of view within an imaginary cylinder extending from the bottom up to the water surface and having a radius of 7.5 m. Record all species observed during the first 5 minutes. To simplify data collection, abbreviate each species name by using the first two letters of the genus and species names (e.g., the Nassau grouper, *Epinephelus striatus*, would be EPST), or the first three letters, if necessary to distinguish between similar names.



During this initial 5-minute period, list only the different species you observe within the cylinder. Do not record data on fish size or numbers of individuals -- except for species that are moving through the cylinder and unlikely to remain there, e.g., sharks, rays, mackerels, jacks.

- 3) **Record number and size of each species on the list.** When the initial 5-minute period is over, begin recording data on the size and abundance of the species you have listed.
- ▶ Working up from the bottom of your species list, count and measure the number of fish of each species, one species at a time, rotating at the 7.5-m mark until the entire area is scanned.
  - ▶ When large schools are present, the number of fish may be estimated by counting by 10's, 20's, 50's, or even 100's.
  - ▶ To estimate fish fork length (from the tip of the upper jaw to the end of the middle caudal rays), compare it to a ruler. Divide each species into size classes based on the minimum and maximum fork length to the nearest cm. The number of classes may depend on the number of fish and the extent of their variation in size, but generally each class should include a size range of no more than 5cm.

- ▶ If a species listed during the initial 5-minute sampling period is no longer present, record data from memory.
- ▶ Any additional fish species observed in the sampling cylinder after the initial 5-minute listing period are ignored unless you want to include them on a site species list.

Each census should take no longer than 15 minutes, including the time needed to record depth, relief of the site, and percent coral cover, sand, and algae. Your underwater data record may look something like the sample shown below, listing the species and the number of individuals of that species counted in each size category. In this example, the observer saw one *Epinephelis striatus* 15 cm long and two 5-7 cm long.

Sample Fish Census Data Sheet			
Species	No. and length of fish by size category		
CHCY	3 (1-2)	56 (3-4)	101 (5-7)
HAAU	2 (10-12)	16 (16-19)	
EPST	2 (5-7)	1 (15)	
GYCI	1 (300)		
CARO	3 (3-4)		

When you enter the data into a computer spreadsheet, record the number of each species and the minimum, maximum and average size of fish in each size class.

**Modifications:** Some investigators have found a smaller radius is preferable in locations where visibility is low. However, the same radius should be used in all surveys to allow for comparability of data over time and among locations; using a modification at your site will make it difficult to compare data with other areas using the standard method.

One modification reduces the cylinder radius to 5 meters and includes any species observed during the entire 15-minute period (Kimmel, 1993). Because many fish adapt to the presence of a diver during the 15-minute period, this modification results in inclusion of more small, cryptic, and sedentary fish (e.g., gobies, blennies, morays). This modified method may also yield better density estimates for small, abundant territorial species.

#### References

Bohnsack, J.A., Bannerot, S.P. (1986) "A stationary visual census technique for quantitatively assessing community structure of coral reef fishes," NOAA Technical Report NMFS 41: 1-15.

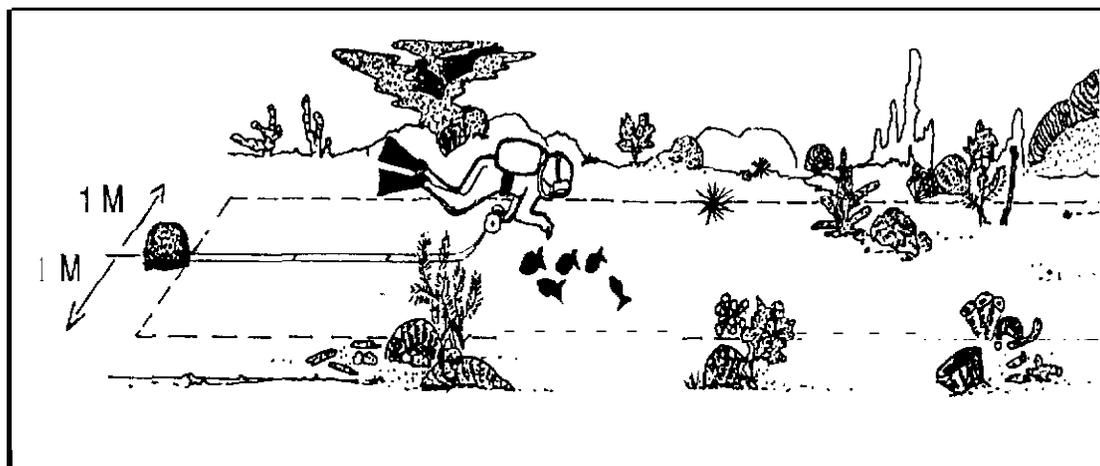
Kimmel, J.J. (1993) "Suggested modifications to diver-oriented point counts for fishes," Proceedings of the 73rd Annual Meeting, American Society of Ichthyologists and Herpetologists, Austin, Texas.

## Belt Transect Census

Belt transects (Brock, 1954) cover a larger area per census than stationary counts and are considered most useful for counting patchily distributed species. They can be conducted along permanent transects marked with survey stakes or other markers, or along reef transects that are randomly selected each time.

The length and width of a belt transect may vary according to the species targeted for a census, but you must use the same dimensions for all transects sampled. A narrow transect (2 m wide) may be good for small, cryptic species, while a wider transect (4 to 5 m) can be useful for groupers, snappers, and parrotfish. Here's the basic technique:

- 1) Swim at a constant speed along the selected area while stretching a fiberglass measuring tape 50 or 100 meters along the bottom.
- 2) As you swim along the transect and unreel the tape, record the fish species, the number of individuals, and the minimum and maximum lengths of species within a prescribed distance (1 to 5 meters) on either side and above the line, including species that are underneath you or cross in front of you. Do not record fish entering the transect area behind you.



*Belt Transect* Census

Swimming speed must be standardized for repeated censuses; highly mobile species may be over-estimated at slow speeds, while cryptic species may be overlooked at faster speeds.

### Reference

Brock, V.E. (1954) "A preliminary report on a method of estimating reef fish populations," *Journal of Wildlife Management* 18:297-308.

## Random Swim Technique

The random swim technique provides good information on relative abundance and species richness, but not on population density. The entire census period is spent searching for unrecorded fish species rather than recording other data about the fish. To obtain reliable data, replicate sample censuses must be conducted.

Variations of this technique appear in Jones and Thompson (1978), and in Kimmel (1985). The basic technique for a 50-minute census is presented below.

- 1) Begin the census at a random location in the selected reef area.
- 2) The **census** period is divided into five 10-minute intervals. Record the name of each species in the interval in which it is first seen.
- 3) To estimate its abundance, each species is given a score based on the interval within which it is first observed. (More abundant species are likely be recorded in the earlier intervals, and the more cryptic or rare species later on.) Species observed in the first interval receive a score of 5, in **the second 4, and so on.**

Your data sheet may look something like this:

Sample Random Swim Data Sheet				
2/5/94: Upper platform, Coral cover: 21%. Depth: 52'				
0-10 min	11-20 min	21-30 min	31-40 min	41-50 min
GOEV	HARA	POPA	CHAN	EPCR
STPL	HAMA	HOTR	SAVE	LAGO
STDO	HABI	CHCA	OPMA	SETO
SCVI	CHCY	ANVI	scco	
SPRU	ABSA	CAPU		
OCCH	CHMU			
HYPV	STPA			
CARU				
CARO				
Score: 5	Score: 4	Score: 3	Score: 2	Score: 1

### References

Jones, R.S., Thompson, M.J. (1978) "Comparison of Florida reef fish assemblages using a rapid visual technique," *Bulletin of Marine Science* 28: 159-172.

Kimmel, J.J. (1985) "A new species-time method for visual assessment of fishes and its comparison with established methods," *Environmental Biology of Fishes* 12:23-32.

<b>Comparison of Fish Census Methods</b>		
	<b>Advantages</b>	<b>Disadvantages</b>
<b>Stationary Census</b>	Good for relative abundance; allows for large sample sizes in distinct habitats.	Takes longer to tram people; less likely to provide a complete site-species list (unless modified).
<b>Belt Transect</b>	Large area can be sampled per census; can target more mobile species; may provide more accurate density estimates for species such as snappers and groupers.	Fewer samples per unit of time than stationary methods; may not provide data on small habitats.
<b>Random Swim</b>	Mostly likely to provide a complete species list; describes a larger portion of species per sample.	Fewer samples per unit of time; provides less quantitative data and no density estimates.

## **Data Analysis**

You need to consider how you'll analyze the data when you design your fish censusing method. It's usually important to analyze data on frequency of occurrence, abundance, richness, evenness and diversity of species at individual sites and among sites. Data on changes in relative abundance and frequency of occurrence can provide information on population changes for individual species. Changes in average size and structure of size classes of important species can also be evaluated.

Data may be summarized using a data management program such as dBase 4 or Paradox, and then analyzed with a statistical package such as SAS or Minitab. Several statistical techniques are available to analyze the data when you have multiple variables (cluster analysis or detrended correspondence analysis) and test for differences among sites (parametric/non-parametric techniques) or over time (time-series analysis). The references listed below provide explanations of these and other statistical methods.

### References

Johnson, R.A., Wichem, D.W. (1992) Applied *Multivariate Statistical Analysis*, Prentice-Hall, Englewood Cliffs, New Jersey, 642 pages.

Ludwig, J.A., Reynolds, J.F. (1988) *Statistical Ecology: A Primer on Methods and Computing*, John Wiley and Sons, New York, 337 pages.

Green, R.H. (1979) *Sampling Design and Statistical Methods for Environmental Biologists*, John Wiley & Sons, New York, 257 pages.