CARIBBEAN COASTAL MARINE PRODUCTIVITY (CARICOMP)

A COOPERATIVE RESEARCH AND MONITORING NETWORK OF MARINE LABORATORIES, PARKS, AND RESERVES

CARICOMP METHODS MANUAL

LEVELS 1 AND 2

MANUAL OF METHODS FOR MAPPING AND MONITORING OF PHYSICAL AND BIOLOGICAL PARAMETERS IN THE COASTAL ZONE OF THE CARIBBEAN

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INTRODUCTION

Caribbean Coastal Marine Productivity (**CARICOMP**) is a regional scientific program to study land-sea interaction processes. The program, based on a cooperative network of marine laboratories, parks, and reserves, focuses on understanding productivity, structure and function of the three main coastal ecosystems in the Caribbean: mangroves, seagrasses and coral reefs. This manual is designed to support the program by providing standard methods for research.

As the primary productivity of mangrove, seagrass and coral reef ecosystems is the basis of present and potential future yields of shallow water marine resources in the region, a primary goal of CARICOMP is to determine the factors that regulate the variability of organic matter production by these systems. In order to do this the CARICOMP program documents and monitors the distribution, structure, and function of the major coastal ecosystems and the extent to which these attributes are influenced by contact with land.

It has become increasingly apparent that Caribbean coastal ecosystems are degrading because of increasing anthropogenic stresses which are superimposed upon natural local, regional and global trends. The long-term monitoring capability of CARICOMP can provide base-line data on Caribbean marine biodiversity and also document threshold responses of ecosystems to global change including human impact and global climate change. The network serves as an infrastructure for large geographic scale studies in comparative ecology and coordinated observations of regional phenomena such as the mass mortality of *Diadema* or a coral bleaching event. Finally, the program will assist in capacity building within the Caribbean regions and enhance management and conservation by making appropriate data and expertise available.

In response to the need for long-term, region-wide comparative studies of the biodiversity and productivity of Caribbean coastal ecosystems, the CARICOMP (Caribbean Coastal Marine Productivity) program began at workshops held at West Indies Laboratory in St. Croix in 1982 and at Discovery Bay Marine Laboratory in 1985. The scientific and management rationale for a cooperative research network of Caribbean marine laboratories, parks, and reserves was established (Ogden and Gladfelter, 1983; Ogden and Gladfelter, 1986; Ogden, 1987). An international Steering Committee was selected (**APPENDIX III)** which subsequently negotiated a Memorandum of Understanding (MoU) with the directors or administrators of 29 sites in 22 countries specifying the responsibilities of the site to the network and the contribution of the program in equipment and logistical support (**TABLE 1, FIGURE 1**). Each site selected a Site Director with the responsibility for implementing the program.

In 1990, 1992, and 1993 workshops were held to coordinate the program and to draft and fine tune the CARICOMP Level I Methods Manual consisting of a minimum number of observations and simple equipment which would permit all members to participate. A Data Management Centre (DMC) was established at the University of the West Indies, Kingston, Jamaica. Data collected according to these protocols are sent to the DMC for storage and preliminary analysis. The DMC distributes analysed data to the participating sites regularly. An annual program summary will be

published in a regional scientific journals/ and or made available on the World Wide Web (WWW). Raw data will also be made available through CARICOMP's Web Site at http:/www.uwimona.edu.jm/centres/cms/caricomp. In addition to providing centralized data processing and storage the DMC coordinates regional investigations of transient oceanographic, biological, and meteorological phenomena and serves as a clearinghouse for new ideas and methods.

In 1996, papers based on the first three years of CARICOMP data were presented at the 8th International Coral Reef Symposium, held in Panama (Appendix II). In 1998 Site Descriptions, including summaries of local research results, were published for 18 CARICOMP sites.

TABLE I
List of CARICOMP Laboratories, Parks, and NGOs and Site Directors
June, 2000 (Addresses, APPENDIX III)

BAHAMAS	Bahamian Field Station, San Salvador	Kenneth Buchan
BARBADOS	Bellairs Research Institute	Renata Goodridge/Hazel Oxenford
BELIZE	Hol Chan Marine Reserve	Alberto Patt/ James Azueta
BELIZE	Smithsonian Institution, Carrie Bow Cay	John Tschirky/Karen Koltes
BELIZE	Marine Research Centre, University of Belize	Eden García
BERMUDA	BBSR (Bermuda Biological Station for Research)	Robbie Smith/Julie Gunther
BONAIRE	Bonaire Marine Park	Catriona Glendinning
CAYMAN	Department of the Environment, Protection & Conservation Unit	Phillippe Bush/Croy McCoy
COLOMBIA	INVEMAR (Instituto de Investigaciones Marinas y Costeras)	Jaime Garzón-Ferreira
COLOMBIA	CORALÍNA (Corporación para el Desarrollo Sostenible del Archipélago de San Andrés, Providencia y Santa Catalina)	June Marie Mow Robinson
COSTA RICA	CIMAR (Centro de Investigación en Ciencias del Mar y Limnología) University of Costa Rica	Jorge Cortés
CUBA	Academia de Ciencias de Cuba, Instituto de Oceanolgía, Centro de Investigaciones de Ecosistemas Costeros (IO-CIEC-IES)	Raul Gomez Fernandez
CURACAO	Stichting Carmabi, CARMABI Foundation	Leon P.J.J. Pors
DOMINICAN REPUBLIC	CIBIMA (Centro de Investigaciones de Biología Marina), Universidad Autonoma de Santo Domingo	Francisco Geraldes
HAITI	FoProBiM (Foundation pour la Protection de la Biodiversite Marine)	Jean Wiener
HONDURAS	Estación de Investigaciones, Cayos Cochinos	Carlos García-Saez
JAMAICA	Discovery Bay Marine Lab., Centre for Marine Sciences, University of the West Indies	Peter Gayle
JAMAICA	CCAMF (Caribbean Coastal Area Management Foundation)	Ann Haynes-Sutton
MEXICO	CINVESTAV, (Centro de Investigación y de Estudios Avanzados del IPN)	Javier Ramirez
MEXICO	ICMyL (Instituto de Ciencias del Mar y Limnología) Estación Puerto Morelos, Universidad Nacional Autonoma de Mexico	Francisco Ruiz-Renteria
MEXICO	Programa EPOMEX-UAC Estación El-Carmen-UNAM, Universidad Autonoma de Campeche	José Luís Rojas-Galaviz
NICARAGUA	MARENA (Ministerio del Ambiente y Recursos Naturales y del Medio Ambiente)	Joseph Ryan
PANAMA	STRI (Smithsonian Tropical Research Institute)	Héctor Guzmán
PUERTO RICO	Department of Marine Sciences, Universidad de Puerto Rico	Ernesto Weil
SABA	Saba Marine Park, Netherlands Antilles	David Kooistra
ST. LUCIA	CEHI (Caribbean Environmental Health Institute)	Joth Singh
TRINIDAD & TOBAGO	IMA (Institute of Marine Affairs)	Rahanna Juman/ Karlene James-Alexander
VENEZUELA	EDIMAR (Estación de Investigaciones Marina de Margarita) (Fundación La Salle de Ciencias Naturales)	Ramón Varela
VENEZUELA	INTECMAR (Instituto de Tecnología y Ciencias Marinas), (INTECMAR), Universidad Simon Bolivar	David Bone/Aldo Croquer

The CARICOMP network of co-operating marine laboratories, parks, and NGOs attempts comparative analysis of data from a broad spectrum of coastal zones, where the structure and function of the component ecosystems differ and the magnitude of terrestrial influence varies. Such comparisons are possible only if comparable data sets are collected using common

methodology. This Level I Manual of Methods stresses the importance of relatively simple techniques, using readily available equipment that is easily maintained, in order to guarantee frequent, regular, and consistently reliable data collection throughout the region. As the program matures it is anticipated that more sophisticated methods will be added to this manual and the sites will be expanded to include more anthropogenically disturbed sites.

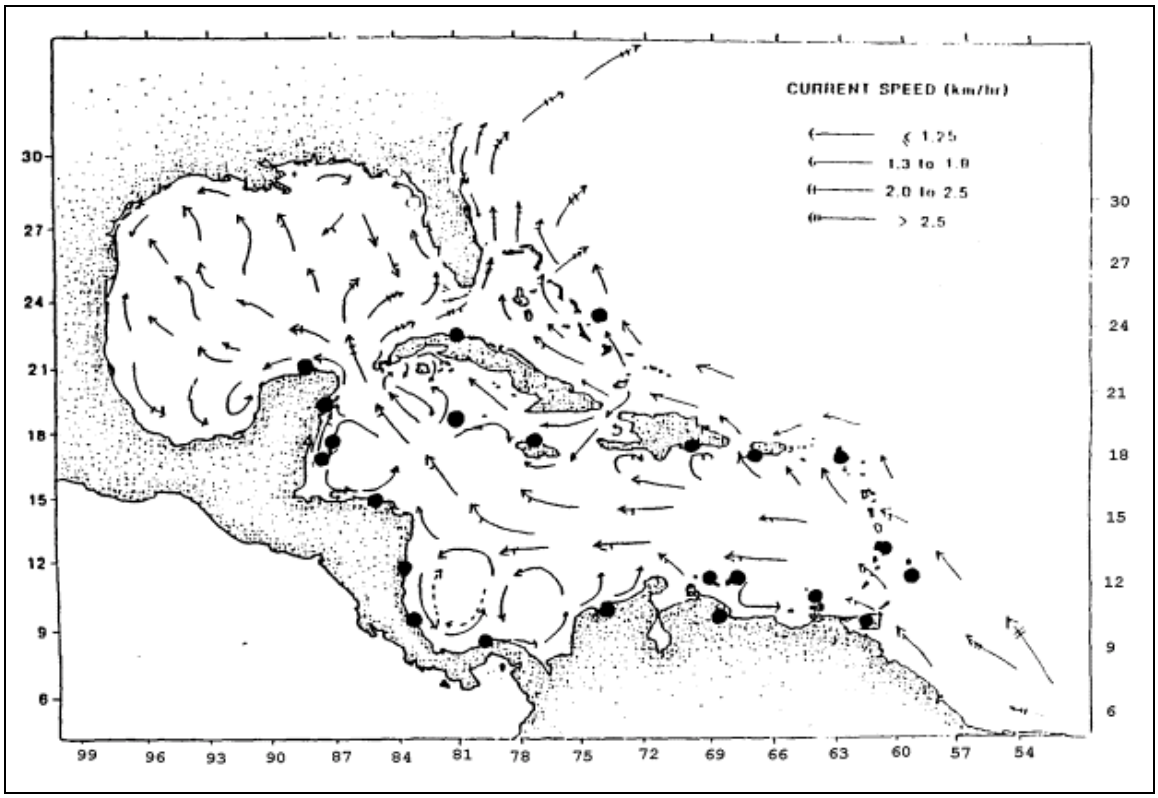

FIGURE 1: Locations of CARICOMP Sites. Arrows indicate speed and direction of the ocean currents interlinking the Caribbean region (after M.J. Shulman and E. Bermingham).

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PHYSICAL MEASUREMENTS

1. INTRODUCTION

The CARICOMP Program includes systematic measurements of a number of physical variables from each of the designated CARICOMP sites. This section of the manual provides guidelines for which physical measurements to make, and how, when, and where to make them.

2. SITE DESCRIPTION AND SELECTION

The research locations at each designated site should be typical representatives of the local environment and include at least one mangrove, one seagrass, and one coral reef habitat. Each habitat should be comparable in size, and function to other habitats of the same type in the local area. Because one objective of CARICOMP is to determine natural community functions, primary stations and transects should be as undisturbed as possible, and removed from anthropogenic perturbations.

Ideally, at each CARICOMP site, sampling should take place along a transect through the three habitats to be investigated: mangrove wetland, seagrass meadow, and coral reef. Preferably the three environmental types should be located adjacent to one another, although it is recognized that at some sites these three habitats will not be contiguous. In such cases, it will be necessary to establish multiple transects or stations that are equivalent to a single transect and are representative of the local habitats and communities.

Information on (or queries about) the location of study sites should be submitted to the Data Management Centre (DMC) for review as soon as sites are chosen.

3. SITE MAPPING

When the optimum locations have been agreed upon for the mangrove, seagrass, and coral reef habitat studies, the Site Director is responsible for mapping and describing the habitats in detail. Mapping should be executed on three separate scales:

1. the basin-wide scale;
2. the landscape scale;
3. the local habitat scale.

A standard legend and a detailed description of the site, landscape, and habitats, relating them to the salient regional and local features will accompany each map.

3a. Basin Map

The basin-wide mapping consists of the placement of all CARICOMP sites on a standard map of the Caribbean basin (approximate scale 1:2,000,000). It is the responsibility of each Site Director to

provide an accompanying description, which will focus on the relationships of the site to the salient regional characteristics, such as climate, meteorology, oceanography, the tectonic and geological setting, geomorphic features, dominant vegetation types, and other features of significance.

3b. Landscape Map

The landscape map scale will vary from one site to the next, depending on the local situation. This map will, if possible, be executed on a scale to include the entire local watershed that impacts on the study habitats at a CARICOMP site. Thus, the scale of this map could vary considerably between the sites, but would ideally be on the scale from 1:50,000 to 1:100,000. This landscape map should indicate clearly the geographic location and relationship between the study habitats; and include scale, latitude and longitude, topographic and bathymetric contours, and standard legends.

The map should be accompanied by a detailed description of the dominant features in the landscape. In the case of an immense watershed, such as the Orinoco, the landscape map may have to be executed as two separate maps in different scales to show both the watershed and the location and relationship between the study habitats. The terrestrial part of the map should be readily available from commercial or government sources. Existing aerial photographs, charts, and satellite imagery may be helpful in mapping marine environments.

3c. Habitat Maps

Detailed maps of the local habitats will be at a uniform 1:5,000 scale for each site. This may necessitate separate maps of each habitat at a site, although a single map showing all three habitats is preferred. The habitat map(s) will show latitude and longitude, scale, water depths, vegetation distribution, sampling locations, and other significant features of the habitat. Each Site Director will produce the habitat map and submit it to the DMC for distribution to other CARICOMP Site Teams.

Suitable maps to provide an outline basis for the habitat maps may already exist. In any case, aerial photographs will be an invaluable source of information, both on land and in the sea where, in clear water, features may be detectable down to 20 m. If such photographs are not already available, the Site Director should seek help in making their own. The camera axis should be as near to vertical as possible, although oblique views can be corrected. Artificial surface marks, the positions of which are known by survey, may be necessary for accurate location of the photographs. It is worth using colour film for the extra information that it yields. Major forest types, sand, seagrasses and reefs are readily distinguishable, but “ground truth” must be determined on foot or in the water. These observers will also compile the habitat descriptions, focusing on local dominant species, environmental processes, and the state of the local environment. It is important that all maps and descriptions should be annually updated. Keep the DMC informed about any changes.

Maps of mangrove areas

Mangrove areas should be mapped to show their geomorphic and physiographic features, and associated water body and channel patterns (Thom, 1967; Pannier and Pannier, 1977; Bacon, 1986); vegetation distribution by species and habitat types (Lugo and Snedaker, 1974); and associated environments. Suggested categories are for features clearly recognizable from aerial photographs,

and in the field, which are of importance to assessing mangrove area structure and productivity. Mapping categories are listed in **TABLE 2**.

TABLE 2. Categories for mangrove area mapping

Rhizophora	- fringe	Salina -	with xerophytic herbs
	- basin	-	unvegetated
	- scrub	Mudflat	
Avicennia	- fringe	Sandbank	
	- basin	Beach	
	- scrub	Channel	
Laguncularia		Lagoon or pond	
Conocarpus		Freshwater source	
Acrostichum		Contours & bathymetry	

Mangrove area habitat maps should be accompanied by verbal descriptions that include details of mangrove habitat types, i.e. fringe, basin, scrub facies; and tabulate total and percentage areal coverage of each. Tabulation should indicate also the total area and percentage area covered by channels, open water bodies, salinas, and areas of degraded mangroves. A summary of the past history of human impacts on the system, e.g. cutting for charcoal and drainage works, should be appended. The location and magnitude of freshwater inputs to the mangrove area should be specified.

Maps of seagrass and coral reef areas

Maps of seagrass and coral reef areas should, if possible, extend to adjacent shorelines and indicate sources of freshwater input. Depth contours should be shown. Major mapping categories are seagrass, sand, hard ground (with or without coral community) and coral reef. Within seagrass and reef areas, dominant species may be useful designators. Among reef areas, reef flat, patch reefs, buttresses and escarpments can be indicated.

Descriptions of the habitat and community types and estimates of their areas will accompany the maps. Known environmental influences will be described, such as freshwater inputs, currents and exposure to wave action.

4. TIME SERIES MEASUREMENTS

Three sets of time series physical measurements will be made (1) daily near the laboratory site (air temperature and cumulative rainfall); (2) weekly on Wednesday (temperature, salinity, and light attenuation) at two of the three habitat sites; and (3) continuous automatic temperature recordings at the seagrass and coral reef transects, in the mangrove wetland, and at or near the ocean and laboratory temperature measurement site. The location of all time series measurement stations should be shown clearly on both the landscape and habitat maps.

4a. Daily Site Measurements

Measurements: Maximum and minimum air temperature (°C) and cumulative rainfall (mm) each 24 hours, recorded once a day together with the temperature at the time of recording the maximum and minimum temperatures.

Measurement Sites: The measurements are to be made at a site near the ocean and near the laboratory. Data from an airport or other established meteorological station within 40 km can be used in lieu of measurements made by the site team. If the measurements are not made locally, the Site Director is responsible for obtaining the substitute meteorological station data.

Measurement Times: Measurements are to be made daily, seven days a week, between the hours of 0700 and 0900 local standard time, LST). Missing values are coded as "MV" if they cannot be measured. Enter instrument values even if they represent more than one day of recording. The time of day that measurements are taken will be entered on the data sheet together with the measured data values.

4b. Weekly Habitat Measurements

Coral Reef

Measurements: Water temperature (°C) at 0.5 m depth. Salinity (psu) at 0.5 m depth. (Note: "psu" is practical salinity unit and is numerically equivalent to parts per thousand, ppt or pro mille). Light attenuation measured vertically with a Secchi disk and expressed as the depth (m) at which the disk no longer can be seen. The location of these measurements must remain fixed from one sampling time to the next.

Measurement Site: Choose a fixed measurement site, adjacent to the studied coral reef, where there is sufficient water depth that the disk disappears from view. If that is impractical, or seems to take you into water of different quality from that at the study site, use the disk horizontally by underwater observation. But be sure to indicate that you have done so on the data sheet. Once you have started making either vertical (preferable) or horizontal Secchi disk measurements at the coral habitat site, you should always continue to make the light attenuation measurements in this manner.

Measurement Time: 1000-1200 local standard time (LST) on Wednesday. In an emergency, it is acceptable to make the measurements on either the preceding Tuesday or subsequent Thursday as long as the measurement times are adhered to. If there is a change in day of sampling from Wednesdays, this should be noted in "Comments" on the data sheet.

Seagrass Bed

Measurements: Water temperature (°C) at 0.5 m depth. Salinity (psu) at 0.5 m water depth. Light attenuation measured horizontally with a Secchi disk and expressed as the distance (m) at which the disk no longer can be seen when viewed through a dive mask 0.5 m below the surface. The site of the seagrass measurements should always remain fixed.

Measurement Site: Choose a measurement site in the seagrass bed habitat where the water depth is at least 1.5 m at the lowest tide.

Measurement Time: 1000-1200 local standard time (LST) on Wednesday. In an emergency, it is acceptable to make the measurements on either the preceding Tuesday or subsequent Thursday as long as the measurement times are adhered to. If there is a change in day of sampling from Wednesdays, this should be noted on the data sheet.

Mangrove Wetland

No weekly habitat measurements need to be made in the mangrove wetland.

4c. Measurement Procedures

Max/Min Air Temperature: Secure a max/min thermometer 1.5 m above the ground, shaded from the sun at all times of the day. Record to the nearest 0.5 °C the maximum and minimum temperature during the past 24 hours, and then reset the thermometer.

Air Temperature at Time of Recording: The actual temperature at the time of max/min air temperature measurements is also read and recorded to the nearest 0.5 ºC from the max/min thermometer, which is secured 1.5 m above the ground and remains shaded from direct sunshine at all times.

Cumulative Rainfall: Secure the rain gauge 1.5 m above the ground in an open area without obstructions within 25 m. Record accumulated rainfall during the past 24 hours to the nearest 1 mm, and then empty the gauge.

Sea Water Temperature: Dip the thermometer into the water 0.5 m below the surface (until the water is up to your elbow) and wait 2 minutes for the thermometer to equilibrate. Record the temperature to the nearest 0.1 ºC.

Salinity: Put a capped bottle into the water 0.5 m below the surface, remove the cap, and let the bottle fill with water, cap the bottle, and return it to the surface. Make the salinity determination in the field by placing a few drops of water on the measurement plate of the temperature-compensated refractometer. Record the salinity to the nearest 1 psu. After making the salinity reading, wash the refractometer with distilled water.

Refractometer should be calibrated annually against known standards. Small deviations from the standard should be included as corrections to the data submitted to DMC. Refractometers deviating by more than 10% from the standard should be returned to the manufacturer for repairs.

Light Attenuation: A standard limnological Secchi disk (0.2 m diameter with black and white pattern) is used to record all light attenuation data.

(i) Vertical: Lower the Secchi disk into the water on the lee side of the boat until it can no longer be seen from the surface (NB: Do not use a dive mask). Record the depth to the nearest 0.1 m. Secchi disk readings must only be made between 1000 and 1400 to be valid, because the sun must be near azimuth.

(ii) Horizontal: Set a permanent pole into the bottom and attach the Secchi disk 0.5 m below the surface with the disk face vertical, and secure the end of a tape measure or a marked rope to the pole. Or, have your buddy hold the disk, and the end of the tape measure or marked rope. Enter the water with a dive mask and swim away from the disk, drawing out the tape measure or marked rope, until the disk disappears from view. Record the observed distance to the nearest 0.1 m.

4d. Continuous Temperature Recording

Measurements: Continuous recording of air and water temperatures (ºC) will be made and logged automatically with a 16-bit Stowaway XTI Data Logger enclosed in a PVC housing sealed with a silicon-greased O-ring.

Measurement Sites: Six temperature loggers/housings have been given to each CARICOMP laboratory. One unit will be placed on the bottom as near to the coral reef transect location as is feasible, where the water depth is 10 m at the time of mean low water; one unit will be placed on the bottom as near the seagrass site as is possible where the water depth is 2 m at mean low water; one unit will be semi-buried in the sediments among the *Rhizophora* prop-roots (in a secure location or suitably anchored to a dive weight) near to the seaward fringe (either subaqueous or subaerial) at the mangrove transect location; one unit will be installed adjacent to the max/min thermometer at an elevation of 1.5 m at the location near to the ocean and the laboratory. The Stowaway XTI Data Logger, enclosed in its sealed housing, must be shaded from direct sunshine at all times of day. The additional two recording temperature units will be used as backup units. Batteries should be replaced in the Stowaway XTI data loggers every 6 – 12 months.

Measurement Times and Programming of Data Logger: The Stowaway XTI data loggers must be programmed prior to field installation using a personal computer with BoxCarPro software, version 3.01 (9/97) or higher (upgrades to v 3.01 are available at http://www.Onsetcomputer.com). Select a sampling interval of 15 mins. and set the "Multiple Sampling" option to "average" (the logger will record the mean of 100 sub-samples taken during the 15 mins. interval. Use "delayed launch" to program the Stowaway logger to launch approximately one hour after it will be deployed in the field (to allow for stabilization of temperature recordings). In preparing the Stowaway dataset to be sent to the DMC, be sure to note those points that should be deleted because they were recorded before or after the data logger was deployed at the measurement site.

4e. Calibration of Temperature Instruments

Frequency of Temperature Calibration: Calibrate and compare all thermometers and Stowaway XTI Temperature Data Loggers against each other at two temperatures once every 12 months.

Calibration of Thermometers: Starting early in the day, fill one ca. 20 litre bucket with ice. Fill the bucket to the brim with fresh water and wait for approximately 1 hour. Put all the thermometers to be calibrated into the ice-water filled bucket. Agitate regularly to ensure the temperature of the water bath is uniform. After one hour, read the temperature of the thermometers without completely taking them out of the ice-water and record the values. During the next hour, read each thermometer every 10 mins. and record the values. Start again by refilling the 20 litre bucket with fresh tap water and put all thermometers into the bucket. Store the water-filled bucket with thermometers at room temperature indoors overnight. Next morning, read the temperature from each thermometer every 10 mins. for one hour and record the values. Report the calibration values to the DMC.

Calibration of Stowaway XTI Temperature Data Loggers: Program the six Stowaway XTI loggers to record data every 10 minutes, beginning on the hour. Place one reference thermometer together with the six loggers (sealed into their housings) in a cooler filled with ice and tap water and leave overnight. The next morning, agitate the water several times and check to see that the thermometer and the loggers are at the same depth in the cooler. Read the thermometer once every 10 minutes for 1.5 hours (10 readings), and record the temperature values and measurement times. Empty the ice water and fill the cooler with tap water at room temperature. Leave the reference thermometer and the six Stowaway units (sealed in their housings) for at least 2 hours, occasionally agitating the bath. Read the thermometer once every 10 minutes for 1.5 hours and record the temperature values

and measurement times. Remove the Stowaway units from the cooler and dry the housings carefully before opening. Download the temperature time series for each logger and determine and record the 20 temperature values that coincide with the times when the reference temperature measurements were made in both the ice-water (10 values) and room temperature (10 values) baths. Report the comparative temperature calibration values for all loggers by serial number (S/N) to the DMC.

CORAL REEF COMMUNITIES

1. COMMUNITY COMPOSITION

1a. Introduction

In any survey, two generalizations apply: (1) the preferred survey method depends on the question being asked, and, (2) sample size sufficiency determines the statistical confidence associated with the survey. CARICOMP studies will be asking two main questions: "is this site different from that one?" and "has this site changed since it was last surveyed?"

Repeat surveys can be of two kinds: repeat surveys of exactly the same location, or repeat surveys of random samples taken from the same general location. Although these two methods may achieve the same goal, the statistical methods and the number of samples required for statistical confidence are different. The random sample method requires many more samples than repeat samples of the same population to detect the same amount of change.

1b. Transects

Coral reefs are complex three-dimensional communities. Sampling methods based on line transects will yield inevitably simplified and distorted information, but they are so simple to use that they remain very useful. Diverse methods have been reviewed (Loya, 1978) and tested, in simulation (Kinzie and Snider, 1978; Ohlhorst et al., 1989) and in the field (Weinberg, 1981; Dodge et al., 1982). Differences of opinion remain on the importance of some details; particularly as measured against the value of time spent underwater, and will be influenced by the specific purpose of each survey. The method proposed for CARICOMP is based on the following criteria:

a. Since CARICOMP is concerned with long-term studies at a number of sites, time spent underwater is not critical.

b. To facilitate comparisons over time, transect sites will be permanently marked and surveyed repeatedly.

c. Since CARICOMP is concerned with productivity, measures estimating surface areas are needed and not merely projected areas.

d. For the same reason, measures of all primary producers, including stalked forms are needed.

e. Overlap among coral reef organisms can be ignored.

f. As with all methods in this Manual, the aim is that it should be replicated easily by different individuals and, as far as possible, be free from observer bias.

The following points are critical in the application of the recommended transect technique:

1. Although each transect should be laid out in a specific depth range, the actual starting points will be randomised.

2. A marker line should be stretched taut along the contour at each station, but the transect line (a chain) should be laid under it, in continuous contact with the substratum. The total length of chain used will be divided by the horizontal distance covered, to give an overall index of the third dimension or **"rugosity"** (Rogers et al., 1982).

3. The continuous intercept method will be used, rather than intermittent points.

1c. Materials required

- 2 x 30 m tape measures, or lines marked at 1 m intervals.
- 1 x 10 m line marked at 1 m intervals.
- 2 x Thin (2-4mm) nylon or dacron white line, about 11-12 m in length, to use as guidelines on the reef transects.
- 1 x Light brass chain (=1 cm link size) a few meters long and marked every 10 **links**. A long chain is hard to handle and awkward to deploy. Most people prefer to use a chain 2-4m in length. Make the chain an even number of links terminating at a 100 point (200, 300, 400 etc.). Thin gauge plastic coated copper wire (from telephone cables) is good for marking the chain with small pieces twisted through a link. The 10, 30, 50, 70, and 90 link positions are marked with one colour, the 20, 40, 60, 80 points marked with another and the 100, 200 etc. points marked with a third colour. Wrap the chain around a reel or place in a Ziploc bag with the starting end hanging out. Make sure the starting end has a short piece of line attached so that it can be tied to the marking stakes.
- 20 x Type 304 stainless stainless stakes; 5/8" (15.9 mm) dia., 24" (61 cm) long. If stainless steel is not available, use steel reinforcing rod.
- **100x Small stainless steel nails, use 1 every m to help to keep chain in place**
- 1 x 2 kg hammer.
- 4 x Star drills; 1/2" (12.7 mm) dia.
- 1 x Underwater compass.
- Underwater writing slates and pencils.

1d. Method

Establishing the 10 transects

1. Choose an area that is representative of the main community expression in that reef area, at a depth of 10 m (+/- 3 m). Try to choose the "best" reef zone, where *Montastraea annularis* is abundant. If this species is not common in the area then choose a zone with high coral diversity. A general reconnaissance may be needed prior to this selection if the observers are unfamiliar with the site.

2. In that community select two separate subareas, that surely belong to the same community expression (separation could be anywhere from a few tens to hundreds of meters, depending on the local variability and the spatial extent of the community).

3. In each of the two subareas place the markers for 5 permanent 10 m length transects. Locate the starting points of the transects randomly. The purpose is twofold: to eliminate subjective bias, and to disperse the transects so that they are truly independent. The methods for randomisation are given below (4-7).

4. Lay a base-line or tape-measure over the reef in a downslope direction from about 1m above the chosen contour depth at your site to 1 m below the contour (e.g. 10 m±1 m or 8 m±1 m, whatever is appropriate for the reef you have chosen; FIGURE 2). Do not let the baseline exceed 29 m in length. Set the base-line to one side of the subjectively chosen area, so that all transect lines will lie on the same side and perpendicular to it (**FIGURE 2**).

5. Use random number tables to choose five points, no less than one metre apart, along the base-line. Those five points will determine the level of each transect, which should begin up to 10 m away from the base-line (**FIGURE 2**). Their exact starting points will be determined by another set of random numbers. You may need to increase the scale of separation between the transects given in the example (constrained within 29 m of transect length) if the "grain size" of the community is large i.e. very large *Montastraea annularis* heads, spurs and grooves.

6. If the slope is very steep, there may not be room for five transects over the two metre depth range. In that case, place two transects to one side of the down slope baseline and three to the other, which should help to keep the transects within the depth range. Try to randomize the determination of the orientation of each transect to the baseline with a coin flip (e.g. heads to left, tails to right), after choosing the random level position (**FIGURE 2**). If the slope is still too steep to accommodate the five transects, then set the transects at random points laterally in a sequence along the chosen depth contour, with two on one side of the baseline and three on the other. Do this by extending the measuring tape away from the random point on the baseline for 40-50 m and choose the random transect starting points on the measuring tape so that the transects do not overlap.

7. Lay out another line or tape measure perpendicular to the base line from the first of the five points chosen above, using an U/W compass, for 30 m. Using the predetermined random number between 1 and 20, locate the beginning and end of the actual 10 m transect.

8. Bore a hole into hard substratum, as near as possible to the chosen starting point, by hammering in a star drill vertically for at least 20 cm. It will be necessary to lift out and rotate the drill from time to time. Then hammer in a stainless steel stake until it jams. Establish the second stake in hard ground also, at least 10 m from the first. Decide from which of the pair of stakes chain transect recording will begin. Label the pair of stakes for each transect with tags or markings, so that there will be no confusion on future surveys. Black and white cable ties make good markers; select colour denoting transect beginning and end and number of ties denoting transect number.

9. Establish the other four transects in a similar manner: then move to the second subarea to establish the other five. **Create a map of the layout of transects to assist in replicating placement of chain each year (note special features which will help in identification).**

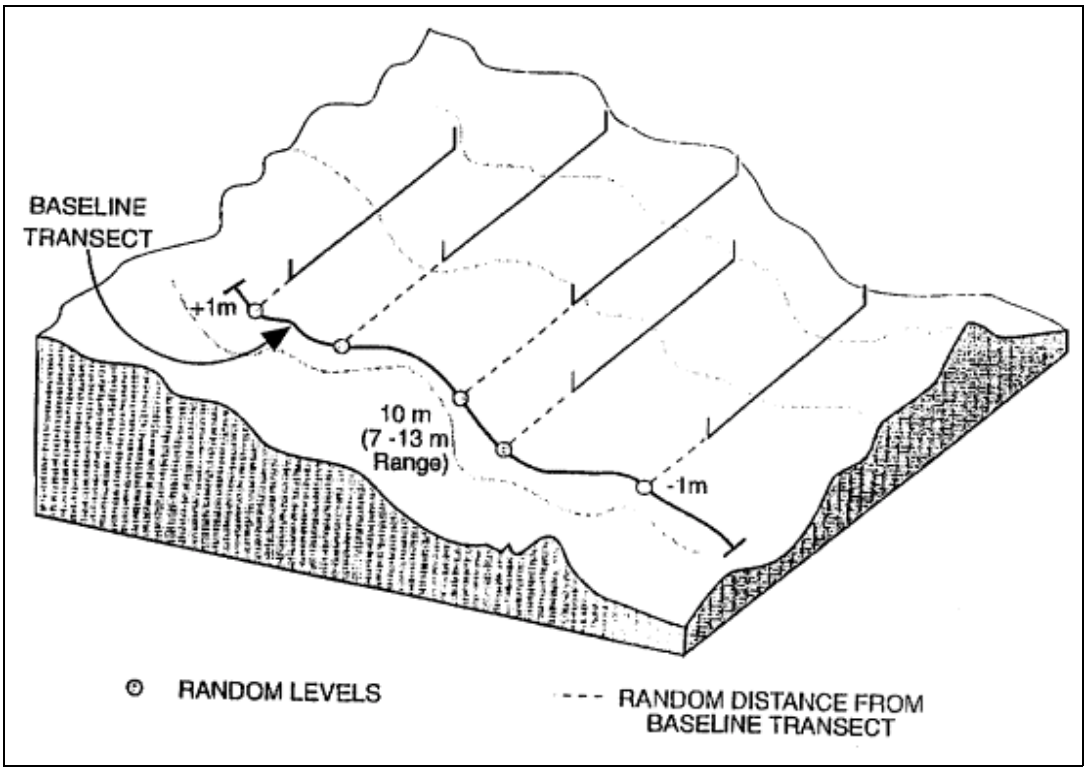

FIGURE 2: Transect Location Procedure

1e. Method

Recording from the transects

1. Secure a thin white nylon or Dacron line (e.g. 2-4 mm heavy fishing line), marked at 1 m intervals and clearly indicating the 10 m point, tautly between the two stakes as a reference line.

2. Lay out the chain on the substratum directly below the taut reference line, from the stake nearer the reference line. It must be adjusted to follow the contour of the substrate, taking care to avoid bridging gaps and crevices. The chain should be laid as precisely as possible, directly below the taut reference line. Checking from above the taut line will help to ensure that the chain is precisely positioned below.

3. On the first occasion (when you are satisfied with the precision of deployment), hammer 2-3" concrete nails into hard ground (try to avoid damage to organisms you will be measuring!) at approximately 1 m intervals along the chain. The nails will mark the position of the chain and allow for more consistent deployments of the chain on future surveys. Place the nails at significant points along the chain, i.e. the top and bottom of large overhangs, where the chain will tend to slip or move

around. If necessary temporarily secure the chain to a nail with a twist of wire or an adjustable cable tie.

4. After the nails are in and the chain has been adjusted to its final configuration, place a marker (e.g. lead fishing weight with a piece of flagging tape) at the position of the last link of the chain. This will be necessary to identify the correct position at which to begin when the chain is moved in order to measure the next section of the transect.

5. After the first chain section has been deployed and the nails and end-marker established, then the successive lengths of substratum components (e.g. coral, rubble, etc.) can be identified, categorized and recorded. The data are collected so as to establish the sequence of components along each transect. To facilitate data collection, set up your dive slate with three columns: the **Link number** at each transition, the **Category** of the previous component, and its taxonomic **identity.** Corals should be identified at least to the genus level and every effort made to identify correctly to the species level. In addition, record whether portions of hard coral measured along transect belong to the same colony.

6. Start at the beginning of the chain and record the unique number that identifies the point of the transition from the first substratum component to the next. If the transition lies between two links, record the last link lying on the first component, not the first link of the next. Write this number down, and record the general category of the component. Identify it to genus and species, but **ONLY** if you are certain. The categories are listed in Table 2. If it is a coral, identify the actual growth form category of the colony under observation, not what its potential future form may be. The general growth forms of coral are shown in **FIGURE 3**. Continue along the chain, recording the last link on each substratum component at the point of transition to the next substratum category. Remember to write down the link number and not the interval or number of links from the previous transition point.

7. Continue along the chain until the end-marker, noting the last transition-point, and then re-deploy the chain. The chain will have to be deployed several times along the transect depending on its length and the rugosity of the reef. When you start to measure the next chain section it will be necessary to refer to the link numbers in a successive sequence from the first chain section. For example, if the first chain stops at 300 links then refer to the next section as Numbers 301-600. It will be less confusing when transcribing the numbers if you write the proper sequence in the field as you record the data.

8. Finish the chain measurements at the 10 m mark on the reference line (which may be before the second stake if the stakes had to be placed more than 10 m apart). Identify the l0 m mark on the substratum with a unique identifier, i.e. a pair of concrete nails set together. This will ensure that future measurements stop at the same point.

9. The rugosity of the transect can be determined from the ratio of the length of chain used to the 10 m length of the transect (a number greater than 1). The standard issue chain has links 1.41 cm long, so rugosity would be given by (total # links x 1.41)/1000.

10. Repeat for the other nine transects! And remember the gorgonian and sea urchin surveys (below).

1f. Notes on recording

Algae

Turf algae may look fleshy and/or filamentous but do not rise more than one cm above the substrate.

Fleshy algae include macroalgae that are not hard to the touch and whose fronds are projected more than one cm above the substratum.

Calcareous algae cover a wide range of species that are generally hard to the touch or pinch. The most common genus is usually *Halimeda*.

Encrusting calcareous algae occur as a hard, smooth pavement on the substratum. They can cover large or small areas. Usually the colour varies from dark pink to purple, and sometimes it may show a greyish hue as well**. Do not confuse it with hard bare substrata that tend to be yellowish or whitish in appearance**.

TABLE 2 Coral Reef Substratum Categories, Codes and Examples

CATEGORY	CODE
Algae	
Turf algae (small fleshy filaments, <1cm high)	TALG
Fleshy Algae (e.g. *Laurencia*, *Caulerpa*, *Sargassum*, *Dictyota*)	FALG
Calcareous algae (e.g. *Halimeda*)	CALG
Encrusting calcareous algae (e.g. *Porolithon*, *Peyssonellia*)	EALG
Hard Corals	
Branching corals (e.g. *Acropora*, *P. porites*)	BRAN
Massive corals (*Montastraea*, *Diploria*, *Dendrogyra*, *Siderastrea*)	MASS
Encrusting corals (*Montastraea*, *Mycetophyllia*, *Agaricia*)	ENCO
Foliaceous (*Agaricia tenuifolia*, *Agaricia sp.*, *Montastraea*, *Leptoseris*)	FOLI
Milleporines (*Millepora sp.*)	MILL
Soft Corals	
Gorgonians (holdfasts, decumbent forms)	GORG
Encrusting gorgonians	ENGR
Anemones	ANEM
Zoanthids	ZOAN
Corallimorpharians	CMOR
Sponges	
Erect sponges	ERSP
Encrusting sponges	ENSP
Other Organisms	
(ascidians, forams)	OTHR
Non-living Substrata	
Bare rock	ROCK
Bare sediment	SAND
Bare rubble	RUBB
Bare boulders	BOUL
Holes, gaps, overhangs	GAPS
Recently dead coral	DCOR

Although these major categories should be recorded, observers are encouraged to provide species level identification if possible. Refer to Field Identification books for help in determining species or category.

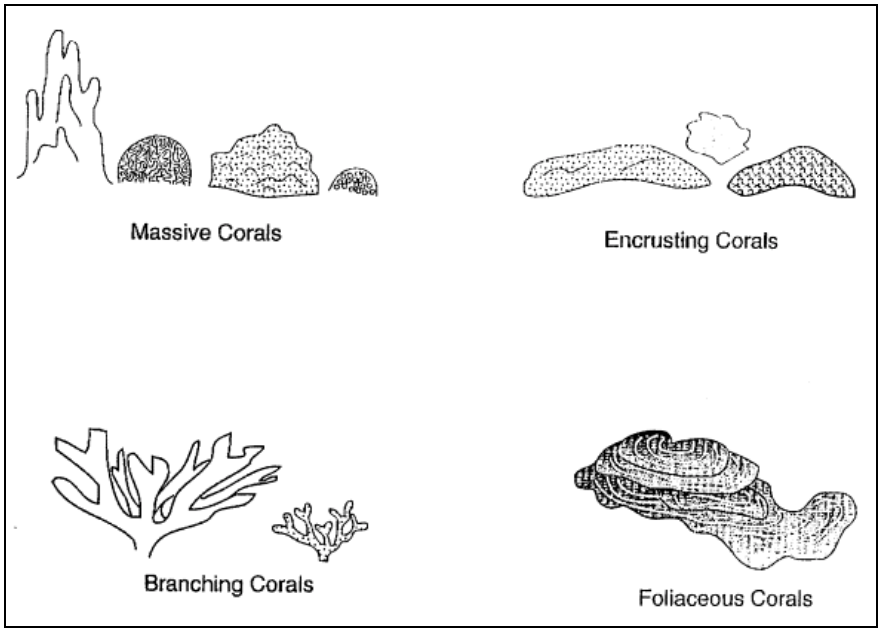

FIGURE 3: Coral Form Categories

Corals

Categorize coral according to the growth form of the colony on the transect, not what it should or could be, i.e. is it encrusting now, not will it be a massive coral when it grows larger. Several species can show a variety of growth forms in different habitats and accurate categorization may be difficult. Consider the entire colony morphology not just the piece or section lying under the transect line (a piece of *M. annularis* may be foliaceous but the entire colony may be better classified as massive). The following criteria will be useful in assigning corals to different categories.

Branching: erect, roughly cylindrical colony that has produced lateral extensions or branches.

Massive: a coral colony that has developed a third dimension such that extends the colony out from the substratum in a mound or domal shape but is unbranched. This would apply to small species such as *Favia fragum* or much larger genera such as *Diploria*, *Montastraea*, *Dendrogyra*, *Siderastrea*.

Encrusting: coral colonies that grow laterally and remain relatively planar in form and conform to the substratum (*Agaricia*, *Mycetophyllia*). Many juvenile or small corals will fall into this category.

Foliaceous: coral colonies that are plate-like or leaf-like and extend off the substratum, projecting into the water (*Agaricia*, sometimes *Montastraea*).

Milleporids: All growth forms of *Millepora* should be included in this category.

The measurement of branched and foliaceous corals can be problematic as there are spaces in between branches and over-lapping sheets. When laying the chain over these colonies do not try to make the chain conform to the indentations and gaps but allow the chain to roughly conform to the overall shape of the colony.

Gorgonians

Be sure the chain lies below any erect colony and is not entangled in the branches. Measure the parts of the colony that are under the chain, which will be primarily holdfasts and low branches. Do not record any detached gorgonians, as they can be considered as temporary occupants of the substratum.

Encrusting gorgonians

The two encrusting gorgonians *Briareum* and *Erythropodium* will be classified as a separate category because of their distinct growth forms, compared to other gorgonians.

Sponges

Encrusting refers to all sponges able to produce a layer over the substrate. Many of these sponges are borers.

Erect sponges are those whose colony is projected above the surface or hangs from it; common examples are tube and vase sponges.

Warning: In many sponges both erect and encrusting, colonial anemones may be abundant. Those are mainly species of the genus *Parazoanthus*, but the morphological attribute to register is still that of the sponge.

Non-living substrata

Bare Rock refers to exposed calcareous substratum or hard ground.

Bare sediment refers to sand (including coarse sand) and mud. Particles have a mean diameter of less than 4.0 mm.

Rubble refers to sedimentary material composed of particles or pieces whose average diameter is 4.0 mm to 30 cm.

Boulders Large blocks of dislodged dead coral heads or terrigenous material, between 0.3-1 m in diameter, that are aggregated and distinct from rubble deposits (<0.3 m).

Holes, gaps and overhangs refer to spaces and voids where it is impractical to measure any of the above mentioned attributes. Overhang refers to the inner space below a projected surface, and also to inner spaces in *Acropora* colonies.

1g. Timing and frequency

Record data from the transects once per year, at the same time each year where possible. It is important that the survey be carried out in calm weather. Strong sea conditions will make the chain deployment difficult, decrease accuracy, and increase the probability that corals and other species on or near the chain will be damaged by the equipment or the divers. We must not bias our results by inflicting damage on the reef communities we want to study over the long-term.

1h. Data Management

Enter the data into the Coral Reef Chain Transect spreadsheets (see Appendices), retaining the exact sequence of link numbers, categories and identifications for each transect. The spreadsheet will calculate the intercept lengths for each substratum category. Make copies of the spreadsheet files and submit them to the DMC.

2. GORGONIAN SURVEY

The number, species and category of gorgonians encountered along each transect at each site will be assessed. After the chain survey is completed, set a measuring tape from the first stake to the second stake of each transect. Swim above the tape-measure and observe any erect gorgonian whose branches or fronds appear to cross above or below the transect line with the normal surge conditions on the reef. Observe all colonies for a 10-15 seconds to see if they satisfy this condition. Stop counting at the 10 m mark of the measuring tape.

Record the position of the holdfast of the colony along the measuring tape (±1-2 cm), and categorize the colony growth form as rod, feather, fan or whip. Finally, identify it to the lowest taxonomic level possible.

Rod: erect, simple or multiple branching or candelabra shape, branches and main axis mostly circular in section, some species have flattened or angular branches.

Feather: Flexible branches with pinnate branchlets in rows (feather-like).

Fan: blade-like fan in one plane or several blades in different planes. Fan conformed by anastomosing branches.

Whip: long thin unbranched stalk.

Record the data on a Gorgonian Survey spreadsheet. Use one spreadsheet for both sites, make a copy and submit it to the DMC.

3. SEA URCHIN SURVEY

The abundance of the long-spined sea urchin, *Diadema antillarum*, will be assessed by swimming a l m long rod or section of PVC pipe along the taut reference line. Use a mark in the centre of the rod to locate the rod over the line so that you will examine an area 50 cm to either side of the line. Take care to look under overhangs and in holes, as the urchins, especially as juveniles, are often cryptic in the daytime.

Data on other urchins, whose abundance may vary inversely with that of *Diadema*, could easily be gathered at the same time. These are likely to include *Echinometra viridis*, *Lytechinus williamsi* and *Tripneustes ventricosus*.

Record the total number of *Diadema* and other urchins encountered along each transect. Enter these data into the Urchins spreadsheet, using one spreadsheet file for both reef sites. Make a copy of the file and submit to the DMC.

SEAGRASS COMMUNITIES

The methods and measurements described below have three general purposes:

to determine the abundance and growth rates of seagrasses at specific sites;
to allow determination of seasonal trends in these measures at the site; and
to allow comparison of the static, dynamic, and seasonal measurements across all CARICOMP sites to develop a true regional picture of the ecology of the Caribbean seagrass communities.

All measures will be expressed on a basis of **grams dry weight per square meter** (g dw/m^2).

1. GENERAL

1a. Sampling Strategy

Probably the most difficult task facing the investigator initially is the selection of appropriate sites for study. Ideally the site should be one that is the most representative of an area. On a practical basis, this can be difficult for even a trained investigator to judge. In Level 1, there will be ideally at least two sites studied, with one site a minimum. The primary site will be the portion of a seagrass bed that visually has the most luxuriant or well-developed *Thalassia* community with clean, green leaves. This site is the simplest to select and will be indicative of the maximum that the area is capable of producing. If possible, a second site will be selected which appears to the investigator to be average and representative of the area in general.

At each site, two stations will be picked for statistical replication. These should be at least ten meters apart, but can be greater distances. They should be visually equivalent.

1b. Timing & Frequency

These Level 1 seagrass samples should be collected twice a year. Because the times of maximum and minimum productivity are not known throughout the CARICOMP network, samples will be taken at times of maximum and minimum day length, which occur in late December and June. As the maximum and minimum day lengths occur late in the months, moving sampling to early January or July is satisfactory. While more frequent sampling is desirable, the processing of these samples requires a great deal of effort. If additional resources are available, it would be best to first increase the frequency of the productivity samples. Additional times of sampling for biomass could then be added later. See Section 5 for a full description of enhanced sampling programs.

2. BIOMASS AND COMMUNITY COMPOSITION OF SEAGRASS BEDS FROM CORE SAMPLES

2a. Introduction

The primary measurements to be made here are the standing crop biomass and total biomass of the plant. For our purposes, the standing crop, or aboveground biomass is composed of the

entire short shoot (i.e. the green and non-green leaves and bundle sheath; see Figure 4a). Leaves will be mainly green, but may be fairly heavily epiphytized. For our purposes here total biomass comprises the standing crop and the belowground, non-photosynthetic portions of the plants. This method for determining seagrass biomass will also yield the seagrass community composition.

The accuracy of the biomass estimate will depend largely on the maximum number of samples that reasonably can be taken and the structural complexity of the seagrass bed. Obviously a better estimate will be obtainable with the same effort for a small monospecific seagrass bed with fairly homogenous shoot density and coverage than for a large mixed-species bed with highly variable shoot densities and coverage.

2b. Equipment

1 ea: Corer; PVC pipe; 30" (77 cm) long, 6-8" (15-20 cm) diameter
A plug, approx. 2-3" (5-7.5 cm) diameter
18" (45 cm) long handle

EITHER
4 ea: Plastic buckets
1 ea: Sieve box; 2 mm mesh
OR
4 ea: Fine mesh (2-4mm) bags (i.e. diving bag)
1 ea: Deep tray
2 ea: Plastic kitchen strainers; 6-8" (15-20 cm) diameter
10 ea: Plastic basins (for sorting different biomass fractions into)
Misc: Aluminium foil
Pink flagging tape
Ziploc plastic freezer bags; quart and gallon size
Hydrochloric or phosphoric acid (10% v/v; 10% concentrated acid + 90% water)
Drying oven (45, 60, or 90 ^{o}C; see text).
Analytical balance.

The best way to obtain biomass samples in Caribbean seagrass beds is by the use of corers. Corers must be sturdy enough to slice through *Thalassia* rhizomes and dense calcareous sediments. Corers may be made from a variety of sources, but those made from polyvinychloride (PVC) pipe are economical and durable. The diameter should be about 15-20 cm and length about 60-80 cm. The cutting end of the corer should be bevelled and notched to provide a better slicing edge. In very compacted sediments, or those with coral rubble, the conventional corer will not penetrate far enough to be useful. In these areas a metal cutting edge needs to be affixed. The simplest material to use is the blade of a band-saw or hacksaw with coarse teeth. This can be affixed on the inner edge of the corer with pop-rivets, and replaced periodically. A continuous handle should go transversely through the corer and be sealed where it passes through the corer barrel. Handles should be at least 15-20 cm on each side for leverage, and be of sufficiently large diameter to be strong and comfortable. The corer must have a removable plug so that a vacuum can be obtained upon extraction, or else much of the material will be lost.

2c. Method

Sample collection

Force the corer into the sediment to at least 45-50 cm to obtain over 90% of *Thalassia* rhizomes and roots. If possible, the corer should be sunk up to the handles so that the sample does not slip down in the corer when removed from the sediments. Important: do not try to just push the corer into the sediments. It must be rotated rapidly back and forth to cut its way into the sediments as it is pushed. Cut - do not just push!

Core samples can be placed in individual buckets. Topside transference of sample from corer to bucket prevents loss of biomass underwater. Alternately, as the cores are taken underwater, they can be immediately extruded into fine (2-4mm), prelabeled mesh bags (e.g. diving bags, laundry bags), while still underwater. With this method, each core can be taken, extruded into its individual bag, and all taken to the surface at the same time, thus eliminating many trips to the surface.

The first few times that cores are taken, they should be taken back to the boat and carefully extruded. Then they should be cut in half lengthwise, and carefully inspected to determine that most of the roots are being collected, and the length of the core from the surface to the bottom of the root zone recorded. There should be a zone at the bottom of the core of 5-10 cm with no roots. This will serve as a guide in the future to just how deep the later cores must be taken.

Treatment of samples

Clean the samples completely of sediment, and separate them first into species of seagrasses, fleshy macroalgae and green macroalgae of the order *Caulerpales* that grow from the sediment. Separation of macroalgae into species is at the discretion of the Site Team. It is desirable but not required.

If the samples are in mesh bags, they can be shaken and massaged while still underwater to remove most of the sediment. If these bags are not available, coarse sorting can be done on a sieve box with a mesh of about 2 mm and washed. Size is not critical but a box about 60 by 40 cm, with sides about 8-10 cm (made of standard 1x3 or 1x4 lumber) is quite satisfactory. The screen must retain small pieces of plant matter and all coarse shell material and fragments must be removed by hand. After coarse sorting, fine sorting can be done on the screen, but is often more conveniently done in a tray of water about 10 cm deep. This greatly aids in sorting the fine fragments. While not a perfect guide, live roots and rhizomes *tend to float*, while dead ones *tend to sink*. Live roots are white or very light grey and crisp when squeezed or broken, while dead roots are dark and more flaccid. Short shoots and rhizomes can have both live and dead roots intermixed. Likewise live rhizomes have a whiter, crisp interior, while dead rhizomes are darker, both inside and out, and are less crisp when broken.

The resulting sample must be all organic matter with no contaminating carbonate fragments. Uncleaned samples can be held without disintegration for a day in shade submerged or several days if running seawater is provided over the sample. Cleaned samples can be held likewise, or,

are best held chilled.

Divide all *Thalassia* plants into the following 5 separate fractions for biomass measurements 1) green leaves, 2) non-green leaves and short shoots, 3) live rhizomes, 4) live roots, and 5) dead below ground material (see **Figure 4a**). Note that the green leaves should simply be torn off at the green/white interface (this is usually at the point where the leaves emerge from the bundle sheath, but may be higher up the leaf if the sheath is buried below the sediment; **Figure 4a**). Note also that the "non-green leaves and short shoots" fraction comprises the non-green portions of the leaves and the sheath bundle, and is simply referred to as "short shoots" in the data sheets.

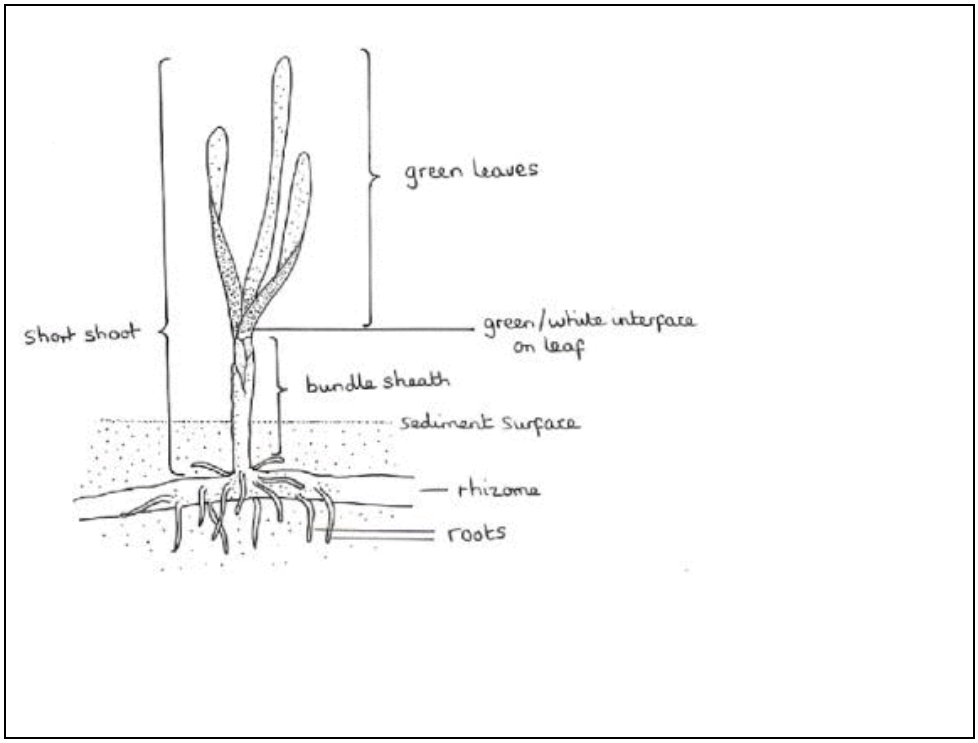

FIGURE 4a: Seagrass plant showing short shoot and components along with rhizome and roots.

For all other grass species, it is generally possible (and often necessary) to simply sort them into 2 separate categories 1) green and 2) non-green tissue.

After separating the plants into their various biomass fractions, make sure that any remaining sediment is removed from the seagrass rhizome and root biomass fractions by scrubbing with a soft toothbrush. Also, epiphytes on the green leaves must be removed in 10% phosphoric (gentler but expensive) or hydrochloric acid (more commonly used and cheap) unless detailed chemical analysis of the components is contemplated. The leaves should be placed in the kitchen strainer and lowered into the acid until bubbling (evolution of CO_2) stops. Note that acid emersion should be as briefly as possible and certainly should not to exceed 5 minutes. The acid

bath must be changed periodically as it becomes less effective.

Rinse or soak all biomass fractions thoroughly in freshwater (using the second kitchen strainer) to remove salt and acid. This is greatly helped if the fine sorting is done in a bath of *fresh water*. Then place each fraction on pre-weighed and marked, heavy-duty aluminium foil tares and dry them at 60-90 ºC to constant weight (no more than 60 ºC if chemical analysis is planned). Some below ground fractions may take several days to dry completely. Until one is familiar with the drying time necessary, it is best to periodically weigh several of the heaviest fractions until they show a constant weight for 12 hours. At this point, all the smaller samples should be thoroughly dry. At this time the samples should be placed in a desiccator to cool before weighing. If no desiccator is available, allow the oven and samples to cool to about 45 ºC before weighing. When through weighing, store the dried samples in a plastic bag for at least 6 months in case any errors have been made and reweighing is necessary.

Divide calcareous macroalgae into above and belowground tissue if desired. Remove all sediment and then decalcify in 20% glacial acetic acid. This may take several days. Fleshy macroalgae need to be rinsed in freshwater, dried, and weighed. Separation into species is generally not required, unless there is a clear dominant macroalgal species in the community.

2d. Data Management

Enter the data on the Seagrass Biomass Data Forms provided by the DMC. To convert the weights per core sample to weights per square meter, multiply by a factor (f) based on the area of the cores. Example: If the area of the core is 200 cm^2, multiply by 50 to obtain data on a square meter (10,000 cm^2) basis.

3. MEASUREMENT OF GROWTH OF *Thalassia testudinum*

3a. Introduction

While the standing crop and biomass measurements will be made on the entire representative seagrass community, initial growth and productivity measurements will only be made on *Thalassia*. *Thalassia testudinum* is the dominant Caribbean seagrass species as it typically contributes more biomass, and thus areal productivity, to total seagrass bed production than *Syringodium filiforme*, *Halodule wrightii*, and *Halophila* spp., and also because *Thalassia* is the competitively dominant and "climax" species in Caribbean seagrass succession.

While standing crop and biomass measurements provide a *static* measure of the condition of the plants at a point in time, the productivity measurements give a *dynamic* measure of the health and growth rate of the plants. In addition to areal productivity (the growth per square meter of sea bottom per unit time), the method allows the determination of the turnover rate of the plants, a measure of the growth per unit weight of the plant.

Growth is measured as the production of new leaf biomass. Production of roots and rhizomes is extremely difficult to measure in tropical seagrass beds and for this reason has rarely been

attempted. Measurement of leaf growth alone will serve the purposes of CARICOMP, although it will not provide an estimate of total seagrass growth.

3b. Equipment

Quadrats: 6 per station marked. Typically 10x20 cm; see text also.
Identification labels for quadrats (see text). Typically engineering flagging tape marked with magic markers.
Hole puncher: today the most commonly used device is a hypodermic needle. Depending on the size of the leaves,
sizes 16, 18, and 21 are most useful.
Scissors (for harvesting shoots after growth)
Plastic Ziploc type bags (the best are Ziploc freezer bags)
Plastic pans to hold acid wash and fresh water wash; typically 3-4.
Plastic kitchen strainer type sieves; 3.
Forceps (to pick seagrass from acid wash)
Pre-weighed receptacles in which to dry seagrass (e.g. aluminium foil, weighing boats, dishes)
Hydrochloric or phosphoric acid (10% v/v; 10% concentrated acid + 90% water)
Drying oven (45, 60, or 90 ^{o}C; see text).
Analytical balance.

3c. Method

Sample collection

At this point, measurements will be made for both productivity and standing crop. The prospective worker is also referred to Zieman (1974) which is both detailed and illustrated, although modifications have been made from the original description (**mainly** the substitution of a needle or hole punch for a stapler).

Although a measure of standing crop was made in the total biomass cores, it is necessary here to measure it *simultaneously* with productivity. Leaf growth should be measured by marking all leaves of a leafy shoot (= leaf bundle = turion = short shoot) a short distance (e.g. 2 mm) above the green-white interface, or at the sediment surface. Commonly the short shoots extend out of the sediments before the leaves divide and become green. The leaves are held tightly together by the bundle sheath and above this point the leaves separate individually, develop chlorophyll, and are green, while below it they are white. It is highly important with this method that all of the leaves on the short shoot be marked simultaneously with a single punch of the needle through all of the leaves.

In many areas, particularly behind reefs or in other high energy areas, the leaf bundle is buried and green leaves emerge from the sediments. In this case the leaves will be both **marked initially** and **collected at a later date** at the sediment surface. When placing the quadrat for marking, sink it to the sediment surface. That way it will serve as a reference point if the sediments are disturbed.

Marked leaves should be left for about 8 to 12 days. The best way to collect the sample is to harvest the entire short shoot from the sediment and return it to the lab intact in a marked Ziploc bag. In the lab, all leaves on a short shoot are clipped at the point on the short shoot at which they were originally marked. This is typically the green-white border that extends closest to the sediment, and is commonly the location of the needle punch mark on the oldest leaf. If the oldest leaf is senescent, indicated by beginning to turn brown or becoming heavily coated with epiphytes, it probably has not grown during the marking period.

In rare circumstances, if the leaves were originally punched individually at the sediment surface, because the sheaths were buried, then they are harvested individually at the sediment surface at this time.

Extinction coefficients (horizontal Secchi disc; Physical Measurements 4.b) for light in the water column above the seagrasses should be measured at least several times (if not daily) during the week when growth is being measured. Two good times are just prior to marking and just prior to collecting, before the divers have stirred up the sediments. Light available for *Thalassia* growth can be calculated using the extinction coefficient and incoming irradiance recorded daily at the site's weather station (see Physical Measurements section).

Laboratory Processing

Leaves and leaf sections are separated into three groups:

Group 1) New Leaves: These are leaves that have emerged since the time of marking. They will be very green and fresh, and are distinguished by having no needle marks.

Group 2) Old Growth (= New growth of old leaves): This group is composed of the length of the leaf from the point of marking down to the base where the leaf was harvested at the original level of marking. It represents the growth of the marked leaves.

Group 3) Old Standing Crop: This is the section of the old, marked leaves *above* the mark. It is a portion of the material that was present when the original material was marked.

Each of these three groups is decalcified in weak acid, thoroughly washed, and dried on an aluminium foil tare. The pieces of foil should have been previously marked for identification and weighed. After drying the total dry weight is measured, the tare weight subtracted, and the actual weight of the plant fraction calculated.

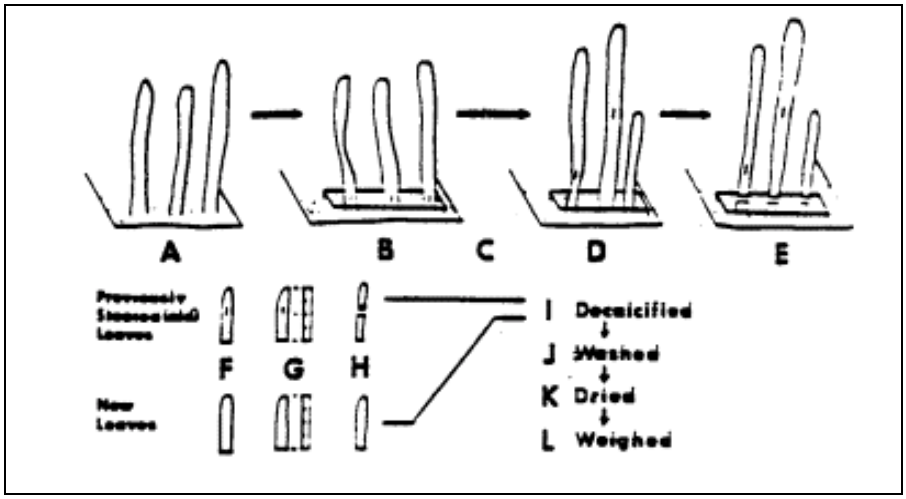

FIGURE 4b: Leaves in a bed (A) are surrounded with a square and perforated (B), allowed to grow (C, D) and harvested (E). The leaves are then divided into old and new blades (F), measured (G), and the new growth removed from the old leaves (H). Finally, the new growth from both groups was decalcified (I), washed (J), dried (K) and weighed (L) (figure from Zieman, 1974).

Areal Productivity is the amount of new material produced per unit area per day. Here it is obtained by summing up the total plant growth (groups #1 + #2) and dividing by the number of days. This figure is the production per quadrat. As a quadrat is 1/50 m^2, this number is multiplied by 50. Thus daily production is defined as:

$$\textbf{Daily Production} = \frac{\textbf{(Weight Group 1 + Weight Group 2)}}{\textbf{\# Days Marked}} \times 50$$

Turnover Rate can be considered in two ways. While areal productivity is the amount of plant produced per unit area, turnover rate is the amount of plant produced per unit plant. Expressed as a percent, it is the percent of the plant present that is replaced each day. Thus:

$$\textbf{Turnover Rate (\%/day)} = \frac{\textbf{Daily Production}}{\textbf{Standing Crop}} \times 100$$

Where Standing Crop is (Gp 1 + Gp 2 + Gp 3) x 50.

3d. Data Management

As before, data (entered on *Thalassia* Growth Forms) should be converted to weights per square meter. Productivity is normally measured in a 200 cm^2 quadrat (1/50 of a m^2), so it is only necessary to multiply all of the values by 50 to obtain the proper value. Thus 50 is the *factor* (f)

for these quadrats.

LEAF AREA INDEX AND LEAF CHEMICAL COMPOSITION

4a. Introduction

A set of measurements will be made to determine leaf area index and several highly useful plant parameters. Leaf area and leaf width are proven indicators of stress in seagrass communities. Both decrease when plants are stressed by such things as excess temperature or salinity. In addition, these measurements will allow better comparisons of the seagrass communities among the different CARICOMP sites across the Caribbean. Plants will also be collected and preserved for analysis of the carbon, nitrogen, and phosphorus content. C:N:P ratios will be calculated which will allow determination of the nutrient status of the plants. These will indicate if nitrogen or phosphorus is limiting or in excess at the CARICOMP sites.

4b. Equipment

Ziploc bags
Small trowel
Scissors / razor blade
Ruler
Drying oven

4c. Method

Sample collection

At the time of leaf marking, the number of short shoots will be counted and recorded for each quadrat. From an area adjacent to the marked quadrats that is visually the same density collect 5 short shoots by uprooting the shoot. Take care not to tear or lose any leaves. Sometimes you will need to dig or fan the sediment away from the base of the shoot with your hand. Place these shoots with their leaves in a separate plastic bag and take to the lab.

Sample processing

At the lab wash the shoots and leaves in fresh water. Cut the leaves from the short shoot with a razor, knife or scissors. Lay them out in the order in which they were on the shoot. Each leaf will then be measured beginning with the youngest, which will be *Leaf 1* to the oldest. Leaf 1 will typically be short, very green with a round tip and in the middle. *Leaf 2* will be the next youngest and will be adjacent to leaf 1. It will *usually* be green with no or few epiphytes but much longer than leaf 1. As the leaves are produced alternately, *Leaf 3* will then be on the opposite side of leaf 1 from leaf 2. *Leaves 4,5* (if present) will continue to alternate from side to side (see diagram).

Measurements

Measure the total length of the leaf from base to tip and record as xx.x cm. Measure the width of the leaf about 1-2 cm from the base and record as xx.x mm. If the leaf is less than 2 cm in length, measure it in the middle. If the leaf still has a round tip record this. Finally measure the length from the base of the leaf to the first occurrence of epiphytes on the leaf and record it in xx.x cm. If epiphytes cover the entire leaf all the way down to the base then record 0.0 for the distance.

After the measurement, use a single edged razor blade or very sharp knife to scrape off all of the epiphytes possible. You may not get all of them and you may scrape off some of the green leaf. This is normal. If the leaves are very yellow and brown and completely covered in epiphytes so that they cannot be reasonably cleaned, or if they fragment so readily that you cannot work with them, throw them away. Rinse them well in *fresh* water, and dry them in the drying oven at **low** temperature (about 40-45°C) until dry. Pack them in a Ziploc bag with a clear label indicating the location from which they were collected and the date that they were collected and send them directly to Jay Zieman (address in Appendix III). They will be analysed for carbon, nitrogen, and phosphorus and the results will be sent to the DMC for distribution back to the CARICOMP network.

5. ENHANCED WORK PLAN

The work plan given above is for the basic Level 1 seagrass work, however numerous investigators have inquired as to what would constitute the logical next step for those interested in additional seagrass studies. This list provides guidelines for additional studies in a coordinated fashion. In general these will involve utilizing the same methods as previous, but intensifying samples both temporally and spatially.

1) **Seasonality**. The next logical step is to increase seasonal coverage at the initial site. Instead of collecting samples twice per year, they would be collected 4 times per year, at 3-month intervals. It is more important to increase this coverage with the productivity samples than with the biomass samples.

2) **Spatial Coverage.** This will involve making a more detailed site map. The area of interest, be a single bed, a lagoon, or a bay, should be gridded off on a map and sample taken throughout the area to determine the **Spatial** variation of seagrasses in the area. For this purpose, the 10x20 quadrats may be used initially instead of the very laborious corers. With a sufficient number of samples, it is possible to obtain a mean density for the area, and to be able to map the areas of high, low, and mean density.

3) **Temporal and Spatial Variation.** With the information obtained in (2), it is then possible to add additional sites that will enable the researchers to capture much more of the local seagrass dynamics. Both productivity and biomass stations should be added. If necessary add the productivity stations first. Biomass may still be done twice per year; productivity can be done twice per year at the new stations, but 4 times per year will be better. Stations can then be located in areas of medium and low density and the more complete variation of the system captured.

MANGROVE COMMUNITIES

General methods for measurement of mangrove ecosystem structure and function are as described by Lugo and Snedaker (1975), Pool et al. (1977), and Snedaker and Snedaker (1984).

The work described below concerns a mangal of red mangroves (*Rhizophora mangle*), fringing the sea coast or a lagoon.

1. COMMUNITY COMPOSITION

Materials required

10 m long tape measure
Sighting Compass, or 4 light 10 m lines
More light line
Stakes, nails and paint for permanent marks
Flagging tape
Large number of weatherproof numbered tags
1 m long cloth tape measure
6 m telescopic measuring rod

1a. Method

Establish 3 plots in a *Rhizophora mangle* forest fringing the coast or lagoon. These will have the letter designations A, B, C, starting at the furthest left-side plot (as seen from the water; **FIGURE 5**). The plots should be identifiable from a permanent marker or by reference to a distinctive feature on adjacent dry land, to aid re-measurement. Other plots could be set up, depending on the size and heterogeneity of the mangrove area being studied (3 to 5 plots).

Plots

Establish 10 x 10 m (0.01 ha) plots at random. Set out each plot by marking a tree at the first corner with flagging tape or paint, measuring a 10 m side, marking the second corner, using the compass to set the second side at right angles to the first, then completing the square in a similar way.

For each plot, all mangrove trees with trunk diameter greater than 2.5 cm should be (a) numbered with a plastic ring-tag or with paint, (b) their position mapped (**FIGURE 6**), (c) identified, and (d) measured as follows:

Diameter - Measure the circumference of the tree in order to obtain diameter of the trunk (which is a standard measure used by foresters; normally expressed as diameter at breast height or dbh). With

Rhizophora the circumference is measured immediately above the buttress roots, using a flexible tape marked in centimetres. Diameter is then calculated as:

dbh = c/pi

It is essential to mark the trunk at the point of measurement (paint a ring or use cord or wire). One year later the tape can be placed on top of this marker when re-measuring the circumference.

Red mangrove trees sometimes have more than one trunk arising from common buttress or "prop-roots." In these cases, each trunk is measured as a separate tree. Where prop-roots grow down from high branches these should be ignored when deciding where to measure the circumference.

Height - should be measured for all trees in the plot using three parameters (a) height above sediment surface of the highest prop root, (b) length of trunk, from prop roots to main area of branching and (c) total height, from ground to highest leaves (**FIGURE 5**). For tall trees a clinometer should be used. For saplings and trees up to 6m a graduated telescoping rod is practicable. **Where tree density is high then measuring height may be very difficult. Estimate as closely as possible, where it may be difficult to obtain actual measurement.**

The measurements to be made are defined below:

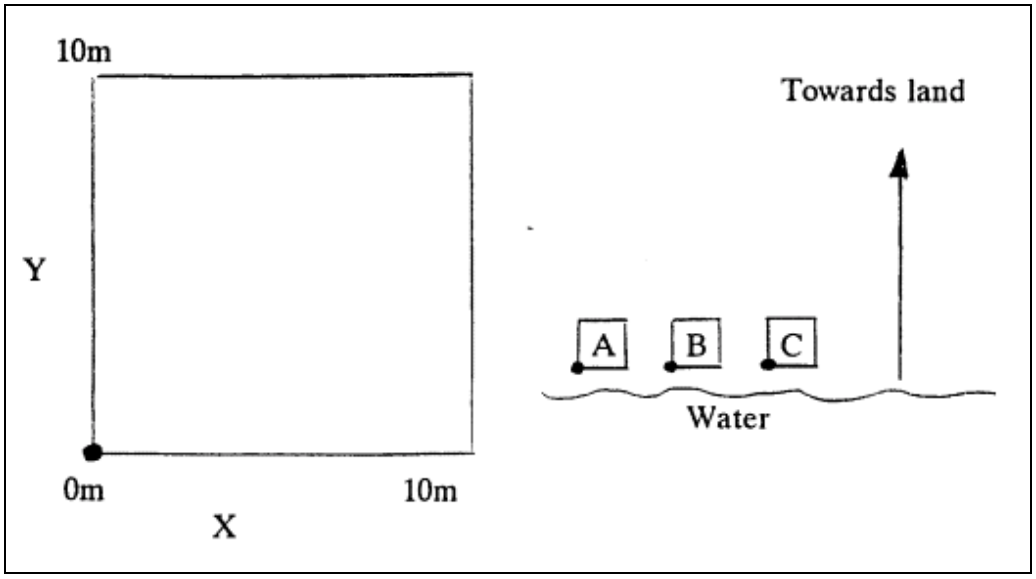

FIGURE 5: Position Mapping; use XY plotting technique within individual plots; the O point in each plot should be at the left side corner closest to the ocean or lagoon, as above.

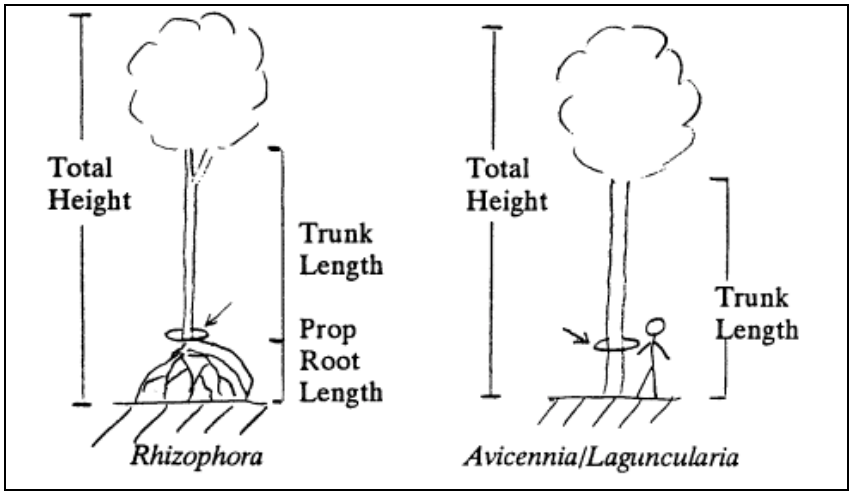

FIGURE 6: Definition of measures to be made in *Rhizophora* and *Avicennia/Laguncularia*. Circle on trunk marks position of circumference measurement.

Sub-plots

Within each plot, establish and mark five randomly placed 1 m^2 sub-plots. In each sub-plot all live saplings (< 2.5 cm dbh) and rooted seedlings should be tagged, identified, mapped and measured as above.

It is recommended that these sub-plots be established and marked before making the tree measurements, then carefully avoid walking in them, to prevent damaged seedlings/saplings from being included in the initial sample.

Note: Some scrub/dwarf mangrove stands may have mature trees with dbh < 2.5 cm. These differ from saplings in fringe and estuarine mangals by the presence of aerial roots arising from branches (in addition to normal prop-roots). For scrub mangroves take the mean dbh of all major stems and record height as above.

1b. Timing and Frequency

Repeat the observations on all trees, saplings and seedlings in the plots and sub-plots at intervals of two years.

1c. Data Management

For comparative purposes, mean diameter will be calculated as the arithmetic mean of all dbh measurements for all plots. The mean stand diameter can be calculated for the whole site mangrove area in which the plots form a sub-sample. Basal area of the stand will also be calculated. This is a measure of the ground space covered by trees, expressed per unit area. For normal trunk forms, *Avicennia* and *Laguncularia*, this is equivalent to the cross sectional area at the point where dbh was

measured. This convention is utilized for *Rhizophora* also, despite the complex prop-root development.

$$\textbf{cross sectional area = basal area = pi x } r^2$$

This will be calculated for each tree in the plot and added together. The total basal area (cm^2) for the plot will be expressed as m^2 ha^{-1} for comparative purposes.

Mean tree height should be recorded for each plot. This, and other measurements can be used for direct comparison with other plots and sites.

Community composition and diversity can be described using the 10x10 m plot data. Comparisons can be made over time (at two yearly intervals), between plots and between sites.

2. INTERSTITIAL WATER

Material Required

Trowel or trenching tool
Suction pump or small container for bailing
Sample bottles

2a. Method

The analysis of surface (tidal) water for temperature and salinity is described in the Physical Methods Section. An important parameter for mangroves is the salinity of the sub-surface water surrounding roots which penetrate the sediments. This is the interstitial water. Its salinity will be measured in each plot.

In areas where surface water is absent, dig a soil pit and remove the soil water by bailing or with the suction pump. Allow the pit to refill with water before sampling. Note the depth of the water table. Where surface water is present, take a small core, using either a cut-off 30-50 ml plastic syringe with the cut edge bevelled to aid insertion, or an equivalent technique. Mark the location (e.g. with a stake) so that the monthly samples will be taken in close proximity to the initial one. The core-hole should be refilled with a core taken from outside of the plot. Where possible, interstitial water in the latter situations should be sampled at low tide. The salinity of the interstitial water should be analysed as described in the Physical Methods section.

2b. Timing & Frequency

Sample interstitial water monthly, preferably at low tide.

3. BIOMASS

3a. Method

Data from the plots and sub-plots are used. In addition, it will be necessary to establish a separate sub-plot from which all saplings and seedlings will be harvested to determine height-weight ratios for juvenile mangrove below 2.5 cm dbh. The sample should contain between 50-100 plants of each species sampled. When harvesting the juvenile plants, use a fork to loosen the sediment around each plant and pull up as much of the root system as possible. **This is done only once to determine the height to weight conversion factor, after which only heights of seedlings are measured in successive samplings**. Data should be expressed as wet weight biomass.

3b. Data Management

Biomass of the mangrove forest trees greater than 2.5 cm dbh is estimated by using trunk diameter and tree density (number of trees per unit area). Individual tree biomass will be calculated using the dbh to weight conversion factor of (1) Golley, et al (1962):

$$\textbf{Biomass (g) = dbh (cm) x 3,390}$$

and (2) Cintron and Novelli (1984):

$$\textbf{Biomass (g)} = b[(\textbf{dbh})^2(\textbf{height})]^m$$

where b and m are constants of 125.9576 and 0.8557, respectively (see paper for details).

Total biomass of trees will be calculated for the plots by summing individual tree measurements. Data should be expressed as wet weight for the living biomass (kg/m^{-2}).

The annual increment in stand biomass due to tree growth is obtained from the differences between measurements of tree dbh made at two yearly intervals.

Biomass of saplings and seedlings from the experimental sub-plot data, using the conversion ratio calculated, will be added to the tree data. Record sapling and seedling growth at two yearly intervals as an indication of recruitment to stand biomass. You should expect some seedling mortality which will influence your estimation of biomass added to the stand.

4. PRODUCTIVITY

4a. Litter Fall (For the fringing *Rhizophora* plots only)

Equipment Required

Litter fall traps, 10 per plot. Construct traps in the form of a basket from a 1 m^2 of nylon netting (mesh size 1.5 mm)
sewn onto a square wood or metal frame 0.5 x 0.5 m in size.
Sample bags, 10 per plot (c. 30 x 20 cm)
0.25 m^2 quadrat frame (0.5 x 0.5 m)

Drying oven
Balance
Wire or string to tie traps
Forceps

4b. Method

Within each sample plot, deploy ten litter fall traps at regular intervals, five parallel to the shore, and five perpendicular to it. The traps should be tied to prop roots of *Rhizophora* approximately one meter above ground, above the highest high tide.

Plot the location of each trap using the xy plotting technique (see **FIGURE 5**). In addition, number each litter trap so that the code for any litter trap will include a letter (for the plot) and a number (for the trap), e.g. A1.

For the first year, litter samples should be collected every month and traps should be left in place throughout the year. Each month, collect litter from each plot (10 traps) into labelled bags and oven dry it at 70 ^{o}C for 48 hours. Divide the litter into leaves, flowers, fruits, bracts, wood/twigs and frass (faeces from herbivore grazing, etc.). Each fraction should then be weighed.

Simultaneously with the first period of litter collection, loose surface litter on the forest floor should be collected from ten 0.25 m^2 quadrats per plot. Bag separately, and label as surface litter from near an identified litter trap (e.g. A1). Floor litter samples should be washed carefully to remove sediment and salt and then dried, sorted and weighed like the trap litter samples.

4c. Timing & Frequency

Surface litter is collected only once, at the start of the survey. Subsequent litter fall collections using traps should be made monthly during the first year. From Year 2, sample four times per year at periods of maximum and minimum production (normally summer & winter or wet & dry season). That is, sample for two months at each period. Eventually trap sampling may be reduced to twice per year, again at periods of maximum and minimum production and traps should be set for one month.

4d. Data Management

Enter surface litter data in the Initial Surface Litter spreadsheet, and monthly litter fall data in the Monthly Litter fall spreadsheet. **Litter fall rates** and **turnover rates** will be calculated by the DMC as follows:

Litter fall rate will be calculated in g m^{-2} d^{-1} and presented graphically, keeping components separate. For interpretation of seasonal changes, graphs of litter fall components can be placed beside meteorological and hydrologic data. Total litter fall (g m^{-2} yr^{-1}) for each plot and the whole site area will be tabulated.

The difference between total litter fall and standing crop of litter, estimated by surface litter collection (although only at one time of year), is an indication of the fate of the litter in the particular habitat. A high standing crop indicates accumulation, while low standing crop indicates removal. Removal is difficult to interpret as it could be due to in situ decomposition or consumption or to export. Export could be into neighbouring water bodies or into neighbouring zones as discussed below. Studies on decomposition rate (below) can be used to aid interpretation, but the turnover rate of litter should be used simply as an index of the amount of organic material available to other components of the ecosystem.

Turnover rate (K) will be calculated as the ratio of total litter fall (L) over 12 months, to standing crop of litter (Xss), assuming a steady state of input = output and rate of change over time as zero, as:

$$\mathbf{K = L/Xss}$$

APPENDIX I

DATA ENTRY TO THE SPREADSHEETS

The CARICOMP spreadsheets are designed for the entry of only raw data. Formulae have been written in to calculate all derived values. To create paper forms on which to collect data, copy the blanks provided or print your own blank spreadsheets.

Sites will differ in the number of rows necessary for their data: use what you need, but do not try to delete unused area as deletion may affect the formulae.

Where data forms are long, we have locked the headers, so that columns can always be identified.

For our data to be compatible, we must use the same conventions for data entry. Where possible, we have programmed the spreadsheet to enforce uniformity. For instance:

1. Dates thus: 01-Jan-94 (enter only the day and month, and the sheet will enter the current year).

2. Times as 4-digit 24hr clock thus: 07:30 15:00

3. All calculations have been pre-programmed, so that you need enter only the raw data.

4. Latitude and Longitude will be expressed in degrees (in the first cell) and minutes and decimals of a minute, entered into a second cell. Thus Lat N : 17 35.2.

For other entries, the sheets remind you of the units (e.g.: ^{o}C, psu (= ppt), grammes, metres, centimetres).

The Spreadsheets

DAILY: Daily site measurements 0700-0900 LST
WEEKLY: Habitat measurements, Wednesday 100-1200 LST
TRAN1-3, TRAN4-5, TRAN6-8, TRAN9-10: Coral Reef Chain Transects

To keep the files small enough to be handled even by a 286, we have allocated no more than 3 transects to each file. So there are 4 files to cover the two groups of 5 transects.

When recording substratum categories on the coral reef transects, we suggest that you use, the same system of 4-letter codes that can be entered in the spreadsheet.

Substratum categories and codes (TALG, BRAN, GORG, etc.) are listed in the Manual (Table 2) and should always be entered. If you are not certain of an organism's name, leave the Species cell blank. If you are certain only of the genus, use the first four letters of its name. If you know the species as well, use the initial letter of the genus, and the first three letters of the species name.

We have included a comprehensive dictionary of categories and taxa in the Coral Reef transect spreadsheets, which will allow the full name of the identified species of corals, sponges and algae to be documented in the template. To save time in data entry and to reduce errors from incorrect spelling, a code system was developed, such that on entering a code in the spreadsheet, if it is in the dictionary, the corresponding full name is entered in the appropriate column. Thus, MONT (or mont, Mont, etc) will enter as "Montastraea sp." and MANN will enter as "*Montastraea annularis*". If the code is typed incorrectly or if the particular organism is not in the dictionary (this should be a rare occurrence) an error message "ERR" will be displayed. Where there are occurrences of duplication of code between organisms, a system of priority coding has been developed at the Data Management Centre to overcome this. The dictionary, along with the guide to its creation, is attached in Appendix 4.

GORGOS: Coral reef gorgonian transect data

All 10 transects are in the one file, with 50 rows per transect. There is no dictionary of names.

URCHINS: Coral reef urchin transect data
BIOMASS: Seagrass biomass data
GROWTH: Thalassia growth data

The six forms are labelled Quadrat 1, etc.

LEAFAREA: Thalassia Leaf Area Index

There are five forms for the five shoots.

FOREST: Mangrove form 1, Forest structure

The first three blocks are labelled Plot A, Plot B, Plot C.

SEEDLING: Mangrove form 2, Seedling/sapling productivity
SEEDBIOM: Mangrove form 3, Seeding and sapling biomass
SURFLITT: Mangrove form 4, Initial surface litter
LITTFALL: Mangrove form 5, Monthly litterfall
INTWATER: Mangrove form 6, Interstitial water samples

APPENDIX II

SUGGESTED READINGS

GENERAL

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APPENDIX IV

CODES USED IN CORAL REEF TRANSECTS

A. Rules for creation of codes for genera and species
B. Examples of the resolution of ambiguities
C. Category codes
D. Dictionary of codes for genera and species

APPENDIX 4a
RULES FOR CREATION OF CODES FOR GENERA & SPECIES

Two simple rules allow the creation of unambiguous 4-letter codes for the majority of Caribbean coral reef benthic genera and species. Use of a few further rules readily resolves the few ambiguous cases.

WHERE BOTH THE GENUS AND SPECIES ARE KNOWN

1st Rule

The 1st letter of the Genus followed by the first three letters of the Species.
e.g. For *Agaricia grahamae* - Agra

2nd Rule

The 1st 2 letters of the Genus followed by the 1st 2 letters of the Species.

3rd Rule

The 1st letter of the Genus followed by the last 3 of the Species.

The coding system attempts to assign unique codes to organisms found on Caribbean reefs in a quick, efficient manner simplifying data entry into spreadsheet templates, such that, arriving at the codes will be intuitive and simple and the organisms can readily be identified by the codes. Ambiguities in coding will arise where the resulting codes for 2 or more organisms are the same. Where ambiguity exists, organisms to be coded are organized in order of priority as indicated below:

1st Priority:	Corals
2nd Priority:	Algae
3rd Priority:	Sponges
4th Priority:	Gorgonians
5th Priority:	Other organisms e.g. zoanthids, corallimorpharians etc.

The Rules of coding will then be applied according to the level of priority given to the

organisms, such that, the 1st Rule is applied followed by the 2nd and the 3rd where necessary.

Example: *Porites porites* (coral) and *Psuedoplexaura porosa* (gorgonian), using 1st Rule both would be PPOR, which is unacceptable. Since *Porites porites* is a coral it takes 1st priority and the 1st Rule applies, whereas the 2nd Rule applies to *Pseudoplexaura porosa*. The new codes are:

Porites porites - PPOR
Psuedoplexaura porosa - PSPO

Where organisms are in the same category e.g. corals, then the more common species will take priority and the priority rules are applied accordingly. Where both organisms may be equally common then order alphabetically.

WHERE ONLY THE GENUS IS KNOWN

1st Rule

The 1st 4 letters of the Genus.
e.g. *Bartholomea* sp. - BART
Montastraea sp. - MONT
Dictyota sp. - DICT

Where ambiguity exists, i.e. 2 or more organisms have the same codes, again the priority system is adapted.

2nd Rule

The 1st 2 letters followed by the last 2 letters of the Genus.

e.g. *Diploria* sp. and *Diplastrella* sp.
Diploria which is a coral takes priority over *Diplastrella* which is a sponge.
Codes are:
Diploria sp. - DIPL
Diplastrella sp. DILA

e.g. the gorgonians *Plexaura* and *Plexaurella* sp. are both quite common. By alphabetical ordering, the 1st Rule is applied to create the code for *Plexaura* and the 2nd Rule for *Plexaurella*, such that the new codes are:

Plexaura - PLEX
Plexaurella - PLLA

A list of ambiguities found in creating codes for the organisms in the dictionary is presented in Appendix 4d.

APPENDIX 4b
EXAMPLES OF THE RESOLUTION OF AMBIGUITIES

	Genus & Species	Category	1st Code	2nd Code	Ordered by
1.	Agaricia grahamae	Coral	AGRA		More Common
	Antipathes gracilis	Coral	AGRA	ANGR	
2.	Amphiroa fragilissima	Algae	AFRA	AMFR	
	Agaricia fragilis	Coral	AFRA		Coral has priority
3.	Antipathes barbadensis	Coral	ABAR		
	Anotrichum barbatum	Algae	ABAR	ANBA	Coral has priority
4.	Botrylloides sp.	Ascidian	BOTR		Alphabetically
	Botryllus sp.	Ascidian	BOTR	BOUS	
5.	Branchiomma nigromaculata	Worm	BNIG	BRNI	
	Botrylloides nigrum	Ascidian	BNIG		Alphabetically
6.	Caulerpa prolifera	Algae	CPRO		Alphabetically
	Cladophora prolifera	Algae	CPRO	CLPR	
7.	Caulerpa serrulata	Algae	CSER		Alphabetically
	Caulerpa sertularoides	Algae	CSER	CASE	
8.	Caryophyllia maculata	Algae	CMAC		Alphabetically
	Caulerpa sertularoides	Algae	CMAC	CSER	
9.	Colangia simplex	Coral	CSIM		Coral has priority
	Canda simplex	Bryozoan	CSIM	CASI	
10.	Cladopsammia sp.	Coral	CLAD		Coral has priority
	Cladophora sp.	Algae	CLAD	CLRA	
11.	Cliona langae	Sponge	CLAN	CLLA	
	Caulerpa lanuginosa	Algae	CLAN		Algae has priority
12.	Dictyota sp.	Algae	DICT		More common species
	Dictyosphaeria sp.	Algae	DICT	DIIA	
	Dictyopteris sp.	Algae	DICT	DIIS	
13.	Dictyota jamaicensis	Algae	DJAM		More common species
	Dictyopteris jamaicensis	Algae	DJAM	DIJA	
14.	Diplastrella sp.	Sponge	DIPL	DILA	Coral has priority
	Diploria sp.	Coral	DIPL		
15.	Galaxaura marginata	Algae	GMAR		Algae has priority
	Gorgonia mariae	Gorgonian	GMAR	GOMA	
16.	Halimeda sp.	Algae	HALI		More common species

	Halisarca sp.	Algae	HALI	HACA	
17.	Isophyllastrea sp.	Coral	ISOP		Alphabetically
	Isophyllia sp.	Coral	ISOP	ISIA	
18.	Mycale laxissima	Sponge	MLAX		Sponge has priority
	Muricea laxa	Gorgonian	MLAX	MULA	
19.	Porites porites	Coral	PPOR		Coral has priority
	Pseudoplexaura porosa	Gorgonian	PPOR	PSPO	
20.	Pseudopterogorgia sp.	Gorgonian	PSEU	PSIA	
	Pseudoplexaura sp.	Gorgonian	PSEU	PSRA	
	Pseudaxinella sp.	Gorgonian	PSEU	PSLA	
	Pseudoceratina sp.	Sponge	PSEU		Sponge has priority
21.	Plexaura sp.	Gorgonian	PLEX		Alphabetically
	Plexaurella sp.	Gorgonian	PLEX	PLLA	
22.	Phyllangia americana	Coral	PAME		Coral has priority
	Pseudopterogorgia americana	Gorgonian	PAME	PSAM	
23.	Polycavernosa crassissima		PCRA	POCR	
	Pseudoceratina crassa	Sponge	PCRA		
24.	Phymanthus crucifer	Anemone	PCRU	PHCR	
	Pseudoplexaura crucis	Gorgonian	PCRU		Gorgonian has priority
25.	Phacelocyathus flos	Coral	PFLO		Gorgonian has priority
	Phyllactis flosculifera	Anemone	PFLO	PHFL	
26.	Pseudocorynactis caribbeorum	Other	PCAR	PSCA	
	Palythoa caribaeorum	Other	PCAR		More common species
27.	Steginoporella magnilabris	Other	SMAG	STMA	
	Sabellastarte magnifica	Other	SMAG		Alphabetically
28.	Siphonodictyon coralliphagum	Sponge	SCOR		Sponge has priority
	Stereotelesto corallina	Gorgonian	SCOR	STCO	
29.	Syringodium filiforme	Algae	SFIL		More common species
	Spyridia filamentosa	Algae	SFIL	SPFI	
30.	Siphonocladus tropicus	Algae	STRO		Alphabetically
	Strongylacidon sp.	Algae	STRO	STON	
31.	Stoichactis helianthus	Other	SHEL	STHE	
	Stichodactyla helianthus	Other	SHEL		Alphabetically

APPENDIX 4c
CARICOMP CATEGORY CODES

Anem	Anemone
Boul	Bare Boulder
Bran	Branching Coral
Calg	Calcareous Algae
Cmor	Corallimorpharian
Dcor	Dead Coral
Ealg	Encrusting Calcareous Algae
Enco	Encrusting Coral
Engr	Encrusting gorgonian
Ensp	Encrusting Sponge
Ersp	Erst Sponge
Falg	Fleshy Algae
Foli	Foliaceous Coral
Gaps	Holes, gaps, or overhang
Gorg	Gorgonian
Mass	Massive Coral
Mill	Milliporine
Othr	Other organism
Rock	Bare Rock
Rubb	Bare Rubb
Sand	Bare Sediment
Talg	Turf Algae
Zoan	Zoanthid

APPENDIX 4d
DICTIONARY OF CODES FOR GENERA AND SPECIES

CODE SCIENTIFIC NAME

#	Code	Scientific Name
1	Acan	Acanthophora sp.
2	Aspi	Acanthophora spicifera
3	Acal	Acetabularia calyculus
4	Acre	Acetabularia crenulata
5	Acet	Acetabularia sp.
6	Acer	Acropora cervicornis
7	Apal	Acropora palmata
8	Apro	Acropora prolifera
9	Acro	Acropora sp.
10	Aber	Actinia bermudensis
11	Aele	Actinoporus elegans
12	Aaga	Agaricia agaricites
13	Afra	Agaricia fragilis
14	Agra	Agaricia grahamae
15	Ahum	Agaricia humilis
16	Alam	Agaricia lamarcki
17	Agar	Agaricia sp.
18	Aten	Agaricia tenuifolia
19	Aund	Agaricia undata
20	Acla	Agelas clathrodes
21	Acon	Agelas conifera
22	Adis	Agelas dispar
23	Asch	Agelas schmidti
24	Agel	Agelas sp.
25	Awie	Agelas wiedenmyeri
26	Atag	Aiptasia tagetes
27	Amir	Alicia mirabilis
28	Acom	Amphimedon compressa
29	Abra	Amphiroa brasiliana
30	Amfr	Amphiroa fragilissima
31	Ahan	Amphiroa hancockii
32	Arig	Amphiroa rigida
33	Amph	Amphiroa sp.
34	Atri	Amphiroa tribulus
35	Asal	Anadyomene saldanhae
36	Anad	Anadyomene sp.
37	Aste	Anadyomene stellata
38	Aors	Anamobaea orstedii
39	Anam	Anamobaea sp.
40	Anba	Anotrichum barbatum
41	Akre	Anthopleura krebsi
42	Avar	Anthosigmella varians
43	Aatl	Antipathes atlantica
44	Abar	Antipathes barbadensis

45	Angr	Antipathes gracilis
46	Ahir	Antipathes hirta
47	Alen	Antipathes lenta
48	Apen	Antipathes pennacea
49	Anti	Antipathes sp.
50	Atan	Antipathes tanacetum
51	Aarc	Aplysina archeri
52	Acau	Aplysina cauliformis
53	Afis	Aplysina fistularis
54	Aful	Aplysina fulva
55	Alac	Aplysina lacunosa
56	Aply	Aplysina sp.
57	Anoc	Arachnanthus nocturnus
58	Azeb	Arca zebra
59	Asyd	Ascidia sydneiensis
60	Atax	Asparagopsis taxiformis
61	Asol	Astrangia solitaria
62	Amag	Astropyga magnifica
63	Aasa	Avrainvillea asarifolia
64	Aell	Avrainvillea elliotii
65	Alon	Avrainvillea longicaulis
66	Anig	Avrainvillea nigricans
67	Araw	Avrainvillea rawsonii
68	Avra	Avrainvillea sp.
69	Aypt	Ayptasia sp.
70	Bflo	Balanophyllia floridana
71	Bala	Balanophyllia sp.
72	Bann	Bartholomea annulata
73	Bart	Bartholomea sp.
74	Boer	Batophora oerstedii
75	Bbru	Bispira brunnea
76	Bvar	Bispira variegata
77	Bmon	Bostrychia montagnei
78	Bten	Bostrychia tenella
79	Bnig	Botrylloides nigrum
80	Botr	Botrylloides sp.
81	Bous	Botryllus sp.
82	Bpyr	Botryocladia pyriformis
83	Bsub	Bracebrigdia subsulcata
84	Brni	Branchiomma nigromaculata
85	Basb	Briareum asbestinum
86	Bria	Briareum sp.
87	Bpen	Bryopsis pennata
88	Bplu	Bryopsis plumosa
89	Btri	Bryothamnion triquetrum
90	Bmin	Bugula minima
91	Btur	Bugula turrita
92	Bgra	Bunodosoma granuliferum
93	Ctri	Calliactis tricolor
94	Ccor	Callithamnion cordatum
95	Cpli	Callyspongia plicifera

96	Call	Callyspongia sp.
97	Cvag	Callyspongia vaginalis
98	Cpod	Calyx podatypa
99	Casi	Canda simplex
100	Crii	Carijoa riisei
101	Cflo	Caryophyllia flos
102	Cama	Caryophyllia maculata
103	Cary	Caryophyllia sp.
104	Ccup	Caulerpa cupressoides
105	Clan	Caulerpa lanuginosa
106	Cmex	Caulerpa mexicana
107	Cpas	Caulerpa paspaloides
108	Capr	Caulerpa prolifera
109	Cpro	Caulerpa prolifera
110	Crac	Caulerpa racemosa
111	Cser	Caulerpa serrulata
112	Case	Caulerpa sertularioides
113	Caul	Caulerpa sp.
114	Ctax	Caulerpa taxifolia
115	Cver	Caulerpa verticillata
116	Cden	Caulibugula dendrograpta
117	Ccla	Centroceras clavulatum
118	Cent	Centroceras sp
119	Cnit	Ceramium nitens
120	Cera	Ceramium sp.
121	Caer	Chaetomorpha aerea
122	Ccra	Chaetomorpha crassa
123	Clin	Chaetomorpha linum
124	Chae	Chaetomorpha sp.
125	Cpen	Chamaedoris peniculum
126	Cpar	Champia parvula
127	Csal	Champia salicornioides
128	Cmin	Chnoospora minima
129	Clit	Chondria littoralis
130	Cten	Chondria tenuissima
131	Cnuc	Chondrilla nucula
132	Chon	Chondrilla sp.
133	Chry	Chrysymenia sp.
134	Cina	Cinachyra sp.
135	Cleu	Cirrhipathes leutkeni
136	Clut	Cladocephalus luteofuscus
137	Carb	Cladocora arbuscula
138	Cdeb	Cladocora debilis
139	Clpr	Cladophora prolifera
140	Clra	Cladophora sp.
141	Cmac	Cladophoropsis macromeres
142	Clad	Cladopsammia sp.
143	Cocc	Cladosiphon occidentalis
144	Clat	Clathria sp.
145	Ccan	Clathrina canariensis
146	Cpic	Clavelina picta

147	Cpue	Clavelina puertosecensis
148	Clav	Clavelina sp.
149	Ccar	Cliona caribbaea
150	Cdel	Cliona delitrix
151	Clla	Cliona langae
152	Clio	Cliona sp.
153	Csub	Clypeaster subdepressus
154	Cdec	Codium decorticatum
155	Cint	Codium intertextum
156	Cist	Codium isthmocladum
157	Crep	Codium repens
158	Codi	Codium sp.
159	Cirr	Coelothrix irregularis
160	Coen	Coenocyathus sp.
161	Cimm	Colangia immersa
162	Csim	Colangia simplex
163	Cola	Colangia sp.
164	Csin	Colpomenia sinuosa
165	Cbre	Colpophyllia breviserialis
166	Cnat	Colpophyllia natans
167	Colp	Colpophyllia sp.
168	Cgig	Condylactis gigantea
169	Cond	Condylactis sp.
170	Cmag	Contarinia magdae
171	Crib	Cribochacina sp.
172	Cvas	Cribrochalina vasculum
173	Cbar	Cymopolia barbata
174	Cmyr	Cystoseira myrica
175	Dbai	Dasya baillouviana
176	Dhar	Dasya harveyi
177	Dver	Dasycladus vermicularis
178	Drub	Davidaster rubinogosa
179	Dcyl	Dendrogyra cylindrus
180	Dost	Derbesia osterhoutii
181	Derb	Derbesia sp.
182	Drii	Desmophyllum riisei
183	Dant	Diadema antillarum
184	Dich	Dichocoenia sp.
185	Dste	Dichocoenia stellaris
186	Dsto	Dichocoenia stokesii
187	Ddel	Dictyopteris delicatula
188	Dija	Dictyopteris jamaicensis
189	Djol	Dictyopteris jolyana
190	Djus	Dictyopteris justii
191	Diis	Dictyopteris sp.
192	Doce	Dictyosphaeria ocellata
193	Dcav	Dictyosphaeria cavernosa
194	Diia	Dictyosphaeria sp.
195	Dbar	Dictyota bartayresii
196	Dcer	Dictyota cervicornis
197	Dcil	Dictyota ciliolata

198	Ddic	Dictyota dichotoma
199	Ddiv	Dictyota divaricata
200	Djam	Dictyota jamaicensis
201	Dlin	Dictyota linearis
202	Dmer	Dictyota mertensii
203	Dict	Dictyota sp.
204	Docc	Dictyurus occidentalis
205	Dcon	Didemnum conchyliatum
206	Dide	Didemnum sp.
207	Dvan	Didemnum vanderhorsti
208	Dsim	Digenia simplex
209	Dnod	Diodogorgia nodulifera
210	Dmeg	Diplastrella megastellata
211	Dila	Diplastrella sp.
212	Dcli	Diploria clivosa
213	Dlab	Diploria labyrinthiformis
214	Dipl	Diploria sp.
215	Dstr	Diploria strigosa
216	Dgla	Diplosoma glandulosum
217	Dcar	Discosoma carlgreni
218	Dneg	Discosoma neglecta
219	Dsan	Discosoma sanctithomae
220	Dber	Distaplia bermudensis
221	Dcor	Distaplia corolla
222	Dcra	Dudresnaya crassa
223	Elun	Echinometra lucunter
224	Evir	Echinometra viridis
225	Etur	Ecteinascidia turbinata
226	Efer	Ectyoplasia ferox
227	Ebar	Ellisella barbadensis
228	Eelo	Ellisella elongata
229	Efle	Enteromorpha flexuosa
230	Ecru	Epicystis crucifer
231	Ever	Ernodesmis verticillata
232	Ebah	Erylus bahamensis
233	Ecar	Erythropodium caribaeorum
234	Eryt	Erythropodium sp.
235	Eisi	Eucheuma isiforme
236	Etri	Eucidaris tribuloides
237	Eobs	Eudistoma obscuratum
238	Eudi	Eudistoma sp.
239	Ecal	Eunicea calyculata
240	Ecla	Eunicea clavigera
241	Efus	Eunicea fusca
242	Elac	Eunicea laciniata
243	Emam	Eunicea mammosa
244	Epal	Eunicea palmeri
245	Epin	Eunicea pinta
246	Euni	Eunicea sp.
247	Esuc	Eunicea succinea
248	Etou	Eunicea tourneforti

249	Eant	Eupogodon antillarum
250	Ecra	Eupolymnia crassicornis
251	Efas	Eusimilia fastigiata
252	Eusm	Eusmilia sp.
253	Ffra	Favia fragum
254	Fhux	Filograna huxleyi
255	Fteg	Flahaultia tegetiformis
256	Gmar	Galaxaura marginata
257	Gobl	Galaxaura oblongata
258	Gala	Galaxaura sp.
259	Gsub	Galaxaura subverticillata
260	Gmin	Gardineria minor
261	Gace	Gelidiella acerosa
262	Gpus	Gelidium pusillum
263	Geli	Gelidium sp.
264	Gnep	Geodia neptuni
265	Gfla	Gorgonia flabellum
266	Goma	Gorgonia mariae
267	Gorg	Gorgonia sp.
268	Gven	Gorgonia ventalina
269	Gcer	Gracilaria cervicornis
270	Gcur	Gracilaria curtissiae
271	Gdam	Gracilaria damaecornis
272	Gdom	Gracilaria domingensis
273	Gmam	Gracilaria mammillaris
274	Grac	Gracilaria sp.
275	Gtik	Gracilaria tikvahiae
276	Gglo	Grifithsia globulifera
277	Gann	Guynia annulata
278	Hrub	Haliclona rubens
279	Hcop	Halimeda copiosa
280	Hdis	Halimeda discoidea
281	Hgor	Halimeda goreaui
282	Hinc	Halimeda incrassata
283	Hlac	Halimeda lacrimosa
284	Hmon	Halimeda monile
285	Hopu	Halimeda opuntia
286	Hali	Halimeda sp.
287	Htun	Halimeda tuna
288	Hsub	Haliptilon subulatum
289	Haca	Halisarca sp.
290	Hbai	Halophila baillonis
291	Hdup	Haloplegma duperreyi
292	Haly	Halymenia sp.
293	Hcuc	Helioseris cucullata
294	Hfer	Hemectyon ferox
295	Heme	Hemectyon sp.
296	Huat	Heterogorgia uatumani
297	Hgib	Heterosiphonia gibbesii
298	Hfee	Hippopodina feegeensis
299	Hhel	Holopsamma helwigi

300	Hcla	Hydroclathrus clathratus
301	Hspo	Hydroides spongicola
302	Hboe	Hydrolithon boergensenii
303	Hcer	Hypnea cervicornis
304	Hmus	Hypnea musciformis
305	Hpen	Hypnogorgia pendula
306	Hyps	Hypsicomus sp.
307	Isch	Iciligorgia schrammi
308	Ibir	Iotrochota birotulata
309	Iotr	Iotrochota sp.
310	Ifel	Ircinia felix
311	Irci	Ircinia sp.
312	Istr	Ircinia strobilina
313	Irig	Isophyllastrea rigida
314	Isop	Isophyllastrea sp.
315	Isin	Isophyllia sinuosa
316	Isia	Isophyllia sp
317	Jadh	Jania adherens
318	Jrub	Jania rubens
319	Jani	Jania sp.
320	Klim	Kallymenia limminghii
321	Lint	Laurencia intricata
322	Lobt	Laurencia obtusa
323	Lpap	Laurencia papillosa
324	Lpoi	Laurencia poitei
325	Laur	Laurencia sp.
326	Lcor	Lebrunia coralligens
327	Ldan	Lebrunia danae
328	Lebr	Lebrunia sp.
329	Lcuc	Leptoseris cucullata
330	Lept	Leptoseris sp.
331	Lasp	Leucandra aspera
332	Lbar	Leucetta barbata
333	Lcer	Liagora ceranoides
334	Lfar	Liagora farinosa
335	Lmuc	Liagora mucosa
336	Lpin	Liagora pinnata
337	Liag	Liagora sp.
338	Lric	Lignella richardii
339	Lcon	Lithophyllum congestum
340	Lobo	Lobophora sp.
341	Lvar	Lobophora variegata
342	Lmed	Loimia medusa
343	Lheb	Lophogorgi hebes
344	Lmin	Lophogorgi miniata
345	Lvir	Lophogorgi virgulata
346	Lyva	Lytechinus variegatus
347	Lwil	Lytechinus williamsi
348	Mdec	Madracis decactis
349	Mfor	Madracis formosa
350	Mmir	Madracis mirabilis

351	Mpha	Madracis pharensis
352	Madr	Madracis sp.
353	Mare	Manicina areolata
354	Mmay	Manicina mayori
355	Mani	Manicina sp.
356	Mpav	Martensia pavonia
357	Mart	Martensia sp
358	Mmea	Meandrina meandrites
359	Mean	Meandrina sp.
360	Mega	Megalomma sp.
361	Msex	Mellita sexiesperforata
362	Memb	Membranipora sp.
363	Mven	Meoma ventricosa
364	Maca	Meristiella acanthocladum
365	Mech	Meristiella echinocarpum
366	Mgel	Meristiella gelidium
367	Meri	Meristiella sp.
368	Mmes	Mesophyllum mesomorphum
369	Mboe	Microdictyon boergesenii
370	Mmar	Microdictyon marinum
371	Micr	Microdictyon sp.
372	Malc	Millepora alcicornis
373	Mcom	Millepora complanata
374	Mill	Millepora sp.
375	Msqu	Millepora squarrosa
376	Mbar	Monanchora barbadensis
377	Mung	Monanchora unguifera
378	Mfav	Montastraea faveolata
379	Mfra	Montastraea franksi
380	Mann	Montastrea annularis
381	Mcav	Montastrea cavernosa
382	Mont	Montastrea sp.
383	Matl	Muricea atlantica
384	Melo	Muricea elongata
385	Mula	Muricea laxa
386	Mmur	Muricea muricata
387	Mpen	Muricea pendula
388	Mpin	Muricea pinnata
389	Muri	Muricea sp.
390	Mfla	Muriceopsis flavida
391	Mpet	Muriceopsis petila
392	Mang	Mussa angulosa
393	Mlae	Mycale laevis
394	Mlax	Mycale laxissima
395	Myca	Mycale sp.
396	Mali	Mycetophyllia aliciae
397	Mdan	Mycetophyllia danaana
398	Mfer	Mycetophyllia ferox
399	Mlam	Mycetophyllia lamarckiana
400	Mree	Mycetophyllia reesi
401	Myce	Mycetophyllia sp.

402	Msty	Myrmekioderma styx
403	Nnol	Neofibularia nolitangere
404	Nspe	Neogoniolithon spectabile
405	Nstr	Neogoniolithon strictum
406	Nann	Neomeris annulata
407	Npor	Neospongodes portoricensis
408	Ngor	Nicella goreaui
409	Nice	Nicella sp.
410	Nare	Niphates areolata
411	Ndig	Niphates digitalis
412	Nere	Niphates erecta
413	Niph	Niphates sp.
414	Nsch	Nitella schmitti
415	Nnud	Notaulax nudicollis
416	Nocc	Notaulax occidentalis
417	Obar	Oceanapia bartschi
418	Osec	Ochtodes secundiramea
419	Odif	Oculina diffusa
420	Orob	Oculina robusta
421	Ocul	Oculina sp.
422	Oten	Oculina tenella
423	Oval	Oculina valenciennesi
424	Ovar	Oculina varicosa
425	Pboe	Padina boergesenii
426	Pgym	Padina gymnospora
427	Pjam	Padina jamaicensis
428	Psan	Padina sanctae-crucis
429	Padi	Padina sp.
430	Pcar	Palythoa caribaeorum
431	Pgra	Palythoa grandis
432	Pmam	Palythoa mammillosa
433	Paly	Palythoa sp.
434	Paca	Pandaros acanthifolium
435	Pdef	Paracyathus defilippi
436	Ppar	Parazoanthus parasiticus
437	Ppue	Parazoanthus puertoricense
438	Pswi	Parazoanthus swiftii
439	Ptun	Parazoanthus tunicans
440	Pcap	Penicillus capitatus
441	Pdum	Penicillus dumetosus
442	Ppyr	Penicillus pyriformis
443	Peni	Penicillus sp.
444	Petr	Petrosia sp.
445	Pebo	Peyssonnelia boergesenii
446	Pcon	Peyssonnelia conchicola
447	Pcri	Peyssonnelia crispata
448	Pina	Peyssonnelia inamoena
449	Psim	Peyssonnelia simulans
450	Peys	Peyssonnelia sp.
451	Psto	Peyssonnelia stoechas
452	Pflo	Phacelocyathus flos

453	Pama	Phorbas amaranthus
454	Pcor	Phormidium corallyticum
455	Phfl	Phyllactis flosculifera
456	Pame	Phyllangia americana
457	Phcr	Phymanthus crucifer
458	Pang	Plakortis angulospiculatus
459	Pfle	Plexaura flexuosa
460	Phom	Plexaura homomalla
461	Pnin	Plexaura nina
462	Plex	Plexaura sp.
463	Pdic	Plexaurella dichotoma
464	Pgri	Plexaurella grisea
465	Pnut	Plexaurella nutans
466	Plla	Plexaurella sp.
467	Ptum	Polyandrocarpa tumida
468	Poly	Polycarpa sp.
469	Pocr	Polycavernosa crassissima
470	Pdeb	Polycavernosa debilis
471	Ppol	Polyphysa polyphysoides
472	Pste	Pomatostegus stellatus
473	Past	Porites astreoides
474	Pbra	Porites branneri
475	Pcol	Porites colonensis
476	Pdiv	Porites divaricata
477	Pfur	Porites furcata
478	Ppor	Porites porites
479	Pori	Porites sp.
480	Ppac	Porolithon pachydermum
481	Poro	Porolithon sp.
482	Prot	Protula sp.
483	Plum	Pseudaxinella lumaecharta
484	Psla	Pseudaxinella sp.
485	Pbip	Pseudo. bipurata
486	Pcra	Pseudoceratina crassa
487	Pseu	Pseudoceratina sp.
488	Psca	Pseudocorynactis caribbeorum
489	Pcru	Pseudoplexaura crucis
490	Pspo	Pseudoplexaura porosa
491	Psra	Pseudoplexaura sp.
492	Pace	Pseudopterogorgia acerosa
493	Psam	Pseudopterogorgia americana
494	Psbi	Pseudopterogorgia bipinnata
495	Pbla	Pseudopterogorgia blanquillensis
496	Peli	Pseudopterogorgia elizabethae
497	Psia	Pseudopterogorgia sp.
498	Pfla	Psuedoplexaura flagellosa
499	Pwag	Psuedoplexaura wagenaari
500	Panc	Pterogorgia anceps
501	Pcit	Pterogorgia citrina
502	Pgua	Pterogorgia guadalupensis
503	Pter	Pterogorgia sp.

504	Ptil	Ptilocaulis sp.
505	Pwal	Ptilocaulis walpersi
506	Reve	Reteporellina evelinae
507	Rjun	Rhaphidophlus juniperinus
508	Rven	Rhaphidophlus venosus
509	Rtom	Rhipilia tomentosa
510	Rpho	Rhipocephalus phoenix
511	Rmac	Rhizosmilia maculata
512	Rsan	Rhodactis sanctithomae
513	Rabd	Rhopalaea abdominalis
514	Rflo	Ricordia florida
515	Rico	Ricordia sp.
516	Rint	Rosenvingea intricata
517	Rose	Rosenvingea sp.
518	Smag	Sabellastarte magnifica
519	Salp	Salpa sp.
520	Sflu	Sargassum fluitans
521	Shys	Sargassum hystrix
522	Snat	Sargassum natans
523	Spla	Sargassum platycarpum
524	Spol	Sargassum polyceratium
525	Spte	Sargassum pteropleuron
526	Sarg	Sargassum sp.
527	Svio	Schizoporella violacea
528	Scal	Schizothrix calcicola
529	Schi	Schizothrix sp.
530	Scom	Scinaia complanata
531	Scub	Scolymia cubensis
532	Slac	Scolymia lacera
533	Scol	Scolymia sp.
534	Swel	Scolymia wellsi
535	Scru	Scrupocellaria sp.
536	Ssid	Siderastraea siderea
537	Srad	Siderastrea radians
538	Side	Siderastrea sp.
539	Stro	Siphonocladus tropicus
540	Scor	Siphonodictyon coralliphagum
541	Sbou	Solenastrea bournoni
542	Shya	Solenastrea hyades
543	Sole	Solenastrea sp.
544	Scus	Spheciospongia cuspidifera
545	Sves	Spheciospongia vesparium
546	Spli	Spinosella plicifera
547	Svag	Spinosella vaginalis
548	Scoc	Spirastrella coccinea
549	Sgig	Spirobranchus giganteus
550	Spir	Spirobranchus sp.
551	Soff	Spongia officinalis
552	Sepi	Sporolithon episporum
553	Spfi	Spyridia filamentosa
554	Shyp	Spyridia hypnoides

555	Stma	Steginoporella magnilabris
556	Sint	Stephanocoenia intersepta
557	Smic	Stephanocoenia michelini
558	Stco	Stereotelesto corallina
559	Shel	Stichodactyla helianthus
560	Slut	Stichopathes lutkeni
561	Sthe	Stoichactis helianthus
562	Stoi	Stoichactis sp.
563	Ston	Strongylacidon sp.
564	Sros	Stylaster roseus
565	Szon	Stypopodium zonale
566	Sexs	Swiftia exserta
567	Swif	Swiftia sp.
568	Svir	Symplegma viride
569	Sfil	Syringodium filiforme
570	Tign	Tedania ignis
571	Tfru	Telesto fruticulosa
572	Tele	Telesto sp.
573	Tame	Telmatactis americana
574	Trii	Thalamophyllia riisei
575	Ttes	Thalassia testudinum
576	Tniv	Thesea nivea
577	Tfra	Titanideum frauenfeldii
578	Tber	Titanoderma bermudense
579	Tpro	Titanoderma protopytum
580	Tita	Titanoderma sp.
581	Tavi	Trematooecia aviculifera
582	Ther	Trichogloea herveyi
583	Treq	Trichogloea requienii
584	Tped	Trichogloepsis pedicellata
585	Tsol	Trididemum solidum
586	Tven	Tripneustes ventricosus
587	Taur	Tubastraea aurea
588	Tcoc	Tubastraea coccinea
589	Turb	Turbinaria sp.
590	Ttri	Turbinaria tricostata
591	Ttur	Turbinaria turbinata
592	Ucya	Udotea cyathiformis
593	Ufla	Udotea flabellum
594	Uocc	Udotea occidentalis
595	Udot	Udotea sp.
596	Uwil	Udotea wilsonii
597	Uhis	Ulosa hispida
598	Urue	Ulosa ruetzleri
599	Ufas	Ulva fasciata
600	Ulac	Ulva lactuca
601	Ulva	Ulva sp.
602	Uoxy	Ulvaria oxysperma
603	NoID	Unidentified species
604	Vaeg	Valonia aegagropila
605	Vmac	Valonia macrophysa

606	Valo	Valonia sp.
607	Vutr	Valonia utricularis
608	Vven	Ventricaria ventricosa
609	Verm	Vermiliopsis sp.
610	Vgig	Verongula gigantea
611	Vrig	Verongula rigida
612	Vero	Verongula sp.
613	Vglo	Viatrix globulifera
614	Warg	Wrangelia argus
615	Wpen	Wrangelia penicillata
616	Wran	Wrangelia sp.
617	Wblo	Wrightiella blodgettii
618	Xmut	Xestospongia muta
619	Xest	Xestospongia sp.
620	Zpul	Zoanthus pulchellus
621	Zsoc	Zoanthus sociatus
622	Zoan	Zoanthus sp.

APPENDIX V

NEW LEVEL I AND LEVEL II METHODOLOGIES

CARICOMP REEF FISH PROTOCOL – LEVEL II

Prepared by: Jaime Garzón-Ferreira, Alberto Rodríguez & Ana Fonseca

1 BACKGROUND

For several years and workshops, the CARICOMP group discussed the necessity of including the reef fish community within the components of its monitoring protocol. Finally, during the last Site Directors meeting of June 2000 in San Jose (Costa Rica), the group decided to incorporate reef fish in Level II monitoring and to prepare a protocol based on the method developed recently for the AGRRA (Atlantic and Gulf Rapid Reef Assessment) program. Here we transcribe the detailed description of the AGRRA fish protocol, with minor changes to adapt it for CARICOMP. The full text can be found in the web page of the AGRRA program at http://coral.aoml.noaa.gov/agra/. The AGRRA fish protocol was developed in 1998 during a workshop at the University of Miami with the participation of some 85 reef scientists from 21 countries, including recognized experts in reef fish community assessment such us Peter Sale and Jim Bohnsack. The AGRRA approach includes two distinct survey methods that provide different types of data and should both be applied at each site (AGRRA, 1999). Method I is designed to measure the density and sizes (used for biomass estimations) of selected Caribbean key fish species, such us predators, herbivores, and "indicator" species, many of which are commercially exploited. Method II is designed to give information on species composition, diversity and abundance.

2 EQUIPMENT

1. Under water templates. An underwater slate on which the data template has been accurately transferred is needed. To facilitate working underwater, the slate can be attached to the T-bar (see below). The datasheet is designed for the fish transect surveys and the roving diver census.
2. At least two 30m fiberglass transect lines with a 3 lb. Weight attached at one of each line. Commercially available PVC surveying tapes are suitable for the transect line, or a 30 m nylon cord attached to a home-made reel will work. A clip can be attached to the reel and suspended from the diver's belt, which allows for the tape to deploy freely as the diver swims.
3. A graduated T-bar or other measuring device (for fish community counts). Construct a T-bar using 1" diameter PVC pipe and a T connector available at hardware stores. It has a 60 cm long handle and two equal length arms providing a total width across the top of 1 m. Use PVC electrical tape or paint to create a scale along both arms showing divisions at each 5 cm.

3 FIELD WORK

Fish assessments should be carried out at least in the two CARICOMP reef sites where coral community surveys are being done, and at two depth intervals: 2-6 m and 9-13 m. As stated before, the protocol includes two distinct methods that should both be applied at each site. However, since Method II requires expertise in visually identifying numerous fish species, it is recommended that only those laboratories with persons experienced in Caribbean reef fish identification, implement this survey. It is also recommended that the fish observations be conducted between 10:00 and 14:00 hours if at all possible, when underwater visibility is at a maximum due to overhead sunlight. Many fishes are wary of humans, hence it is necessary to keep away from other people while making these observations.

Method I: Belt transect counts for defined species list

1 Lay a 30m transect line by first placing the weighted end of the line on the bottom, and then swimming in a straight line while releasing it from the reel as you count the fish. This minimizes the disturbance to the fishes prior to their being counted. Periodically fixing on an object in the distance as you swim will help you swim in a straight line. (You can clip the transect tape to your weight belt to allow for easy release of the tape).

2 As you swim out the full 30m transect line, count and record fish found within a 2 m wide visually estimated belt transect. Carry a data sheet in standard format, and a 1-m wide T-bar to ensure accurate monitoring of the 2-m wide belt. Hold the T-bar ahead of you angled downward at about 45 degrees, and try to focus your gaze on the several meters of the transect ahead of the T-bar. Count only those SPECIES listed below and do not count juvenile parrotfishes or grunts less than 5 cm in total length. This list of species has been chosen to provide coverage of a number of the species most likely to be affected by human impacts, while preserving a relatively consistent search image. This should enhance the precision of transect data.

SELECTED FAMILIES- include EVERY SPECIES within the following families:

1. Surgeonfish (e.g., *Acanthurus bahianus, A. chirurgus, A. coeruleus*)
2. Parrotfish (e.g., *Sparisoma viride, S. aurofrenatum, S. rubripinne, Scarus taeniopterus, S. serti*)
3. Grunt (e.g., *Haemulon flavolineatum, H. chrysargyreum, H. sciurus, H. plumieri, H. aurolineatum*)
4. Snapper (e.g., *Lutjanus griseus, L. apodus, L. mahogoni, Ocyurus chrysurus*)
5. Grouper (e.g., *Epinephelus guttatus, E. fulvus, E. cruentatus, E. striatus, Mycteroperca bonaci*)
6. Angelfish (e.g., *Pomacanthus paru, P. arcuatus, Holocanthus tricolor, H. ciliarus*)
7. Butterflyfish (e.g., *Chaetodon capistratus*)

8. Triggerfish (e.g., *Balistes vetula, Melichthys niger*)

ALSO COUNT the following six species:

9. Threespot damselfish (*Stegastes planifrons*)
10. Yellowtail damselfish (*Microspathodon chrysurus*)
11. Hogfish (*Lachnolaimus maximus*)
12. Spanish hogfish (*Bodianus rufus*)
13. Barracuda (*Sphyraena barracuda*)
14. Bar jack (*Caranx ruber*)

3 Estimate the size of each fish and assign them to the following size categories (<5 cm, 5-10, 10-20, 20-30, 30-40, >40 cm) using a 1 m T-bar with 5 cm increments to assist in estimating sizes. Large groups of individuals of a species will be classified by attempting to put them into one or more size categories as necessary. By remembering to keep effort equivalent on all segments of the transect, you can limit the tendency to count all members of a school crossing the transect, instead of just those members which happen to be within the transect as counting of that segment takes place. Record data in the Reef forms which can be found at http://www.reef.org/data/scanforms.htm.

Note: Sample the transect belt giving uniform attention to each successive 2-m segment. This requires swimming at a more or less constant rate, while looking consistently about 2 m ahead of your current position. You may pause while recording data, and then start swimming again. It is important to swim in a uniform manner and a speed that covers each 30-m transect in 6-8 minutes should be attempted. High densities of counted species will slow this rate in some cases. The diver will be tempted to count all members of a school as they swim across the transect unless he/she concentrates on giving equal effort to sampling each successive portion of the transect. Only those school members that are actually within the 2 m wide strip of that segment of the transect at a given time is included in the census. Fish observers should be trained to estimate fish lengths by using consistency training methods both on land and underwater.

1. When you reach the end of the transect line, stop the survey and recoil the transect tape.
2. Continue conducting haphazardly-positioned 30 m transects, at least 5 m laterally away from the previous position, until completing at least 5 transects by each depth interval and site. Repeat the above steps for each transect.

Modifications: Some workers may want to census other species of fish. This is encouraged, provided that these other species are counted on a SEPARATE pass over the transect, after the AGRRA run. Otherwise the census method is substantially changed, and your data may not be directly cross-comparable with other AGRRA assessments.

Method II. Rover Diver census

After finishing the belt transects, conduct a roving diver census of ALL SPECIES of fishes following the methodology of Reef Environmental Education Foundation (REEF) (http://www.reef.org/) and briefly explained below.

- The Rover diver census is conducted in the same general area as the belt transects are set.
- Swim around the reef SITE for approximately 30 minutes (preferably 45-60 min - the longer the better) and record ALL fish species observed. Use all knowledge you have of fish habits, and search under overhangs, in caves, and so on. The objective is to find the maximum number of species that you can during your search time.
- Estimate the density of each species by using logarithmic categories: Single (1 fish), Few (2-10 fishes), Many (11-100 fishes), or Abundant (> 100 fishes).
- Record your observations on the standardized REEF data entry sheet.
- Contact REEF to submit your datasheet for the regional database.

4 REFERENCES

AGRRA. 1999. Atlantic and Gulf Rapid Reef Assessment (AGRRA). Mesoamerican Reef System Workshop. May 17-21, 1999. RSMAS, University of Miami.

CURRENT STATUS OF POPULATIONS OF THE BLACK SEA URCHIN *Diadema antillarum* Philippi IN THE WIDER CARIBBEAN

A wide spread mass mortality in 1983-84 significantly reduced the populations of the black sea urchin *D. antillarum* in the Caribbean. The cause was presumably, a species-specific pathogen. The urchins disappeared completely from many coral reef localities. In some of these localities, where other herbivores were in low numbers or absent, the disappearance of this key herbivore, brought about changes to the algal and coral community structure. Seventeen years have passed since this die-off event happened, and current observation indicate that *D. antillarun* is coming back strongly in many Caribbean localities (Weil, personal observations). The potential of this organism to reach high densities (low number of predators) and produce changes in the algal and/or reef communities makes this project an important contribution. It is necessary to : (1) determine what is the current status of the populations of *Diadema* in the wider Caribbean, and (2) to develop a simple monitoring program to follow the recovery (or not) of this organism. Future research topics will include the assessment of what (if any) changes and at what rates are these changes occurring in different coral reef communities due to the return of *Diadema*. It is also possible that recurrent die-off in isolated localities may occur, or that another mass mortality event will hit this species in the future. In any case, baseline information on the population status and dynamics of this organism will be of great importance.

OBJECTIVES:

The main objectives are: (1) to determine the current status of the populations of *D. antillarum* in the wider Caribbean; and (2) to start with a monitoring program to follow the population dynamics of this urchin over time in several localities across the Caribbean.

METHODS:

Ideally, each Site Director should be collecting data on the DENSITIES (ind/m²) and SIZE STRUCTURE (or at least, proportions of juveniles and adults) at different depth intervals (varying habitat spatial heterogeneity) in at least five local populations (coral reef areas) of *Diadema* in each CARICOMP site. Participant Site Directors should find all historical data and information about *D. antillarum* before and after the mass mortality for his/her localities. A two-level protocol was developed to: firstly, accomplish the major goal of the project, and secondly; for those willing to go the extra-mile, to gather more information on the characteristics and spatial distribution of the sea urchin populations.

LEVEL-1 PROTOCOL

1. Survey, by snorkeling, at least five reef sites in your locality down to 5 meters (0-15 feet) in depth. Count the number of urchins you see in two, 15 minutes of snorkeling per reef. Try to visually separate urchins in at least two size classes, those below 5 cm in test diameter ("juveniles') and those above 5 cm in test diameter (adults). Make observations about the spatial heterogeneity of the habitats and write it down.

2. After you have surveyed at least five reef sites, select at least three reefs (include your CARICOMP site if it has high numbers of *Diadema*). Describe the physiographic characteristics of the reef and determine if it has:
   - a back reef area
   - a front reef area with a slope (down to at least 12-15 m)
   - there are reef sitess, like Bermuda, that have no slope. These should make an effort to survey at least to different areas of the same depth interval.

3. The front reef and slope should be divided into three depth intervals:(0 - 5 m, 5 - 10 m and > 10 m). In each depth interval, at least five, 10 x 2 m, band transects, randomly placed, should be used to count the number of sea urchins and to determine the proportion of juveniles/adults or the size structure of the populations. If your reef has a back reef area with urchins, then do five band transects in this area also.

4. The first transect at each depth interval should be placed haphazardly at a depth in the middle of each interval (i.e. 2.5 m, 7.5 m and 12.5 m). Using a random number table or generator, select two numbers between 0 - 100 for each of the following four transects in each depth interval. If the first number is even, then move the transect up the slope. If the number is uneven, move the band transect down the slope. Keep the transects within the depth interval. Use the second random number as the number of forward fin kicks to move you to the next position, and then, lay down the next transect. Repeat this process for each band-transect.

5. Once your transect line is in place, swing on each side of the line and count all urchins observed within a one meter area from the line. Take a 1 m long pvc section to mark the distance. Try to separate number of urchins under 5 cm of test diameter (juveniles) and those larger than 5 cm in test diameter (adults). Be careful while doping the surveys, look under all ledges, inside holes and crevices, etc., and count all urchins within the 20 m sqm band transect (your sample unit).

6. If the reef has a well developed back-reef area with many urchins, then select this habitat in addition to the front reef habitat and survey 5 band transects.

7. Data should be put in a data sheet that will include, date, locality, site, depth interval, transect number, number of juveniles per transect, number of adults per transect, total number of urchins, proportion of juveniles, proportion of adults, and density per transect.

8. TIMING - sampling should be carried out before the end of the year.

LEVEL-2 PROTOCOL

The level two protocol will provide information on the size structure of the different urchin populations and the relationship between the urchin densities and the size structure to the spatial heterogeneity of the different reef habitats.

1. After finishing with the level-1 density count, determine the rugosity of each transected area using the same method used for the coral reef surveys.

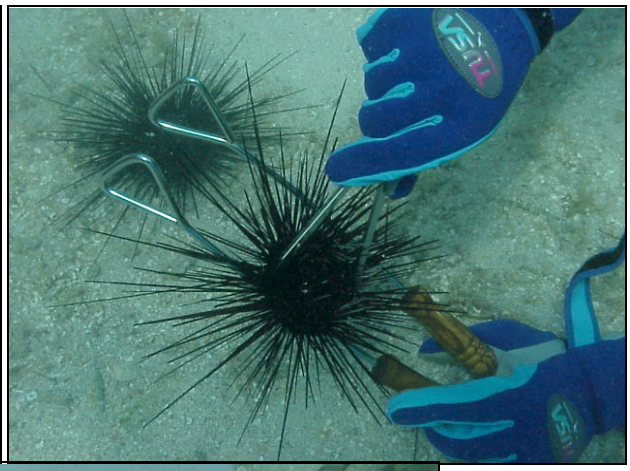
2. Collect at least 200 urchins of all sizes along each depth interval. For this you need a large, plastic basket, barbeque thongs, a barbeque fork and a dive buddy. Move to a clear bottom area. Using the thongs, pick individual urchins from the basket, turn them upside down and then measure the oral test diameter using a two pointed compass. Compass points should be touching the test. Check the open compass distance with a metric scale glued to your UW slate. Write down the diameter of each urchin in cm on your slate. (See photographs). The easiest way is for one person manipulate the urchins and measure them while the other person holds the slate and writes down the measures. Use gloves to prevent prickling yourself to often.

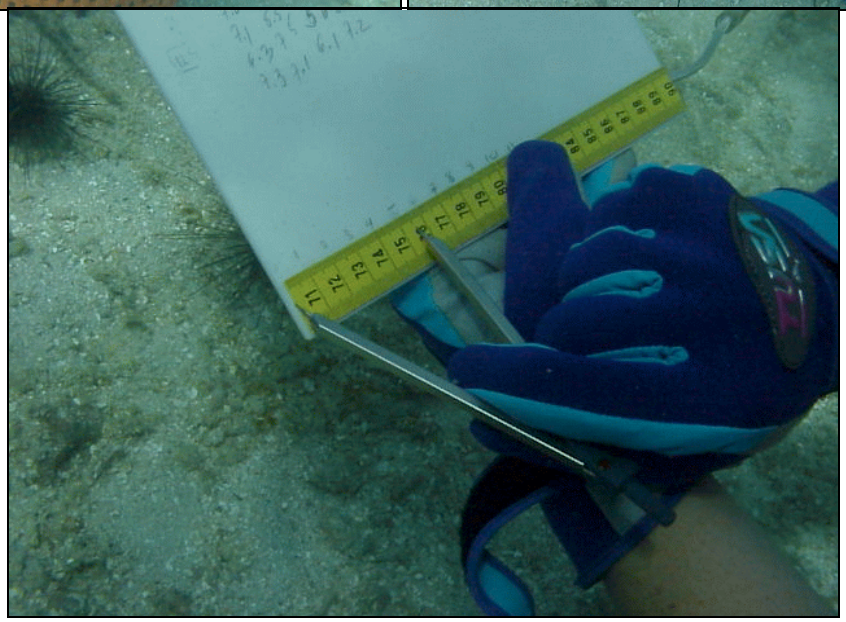
3. Two persons can easily do one or two reefs per day after acquiring a minimum of experience handling the urchins. All density counts can be done in one dive. Collection and measurements will take longer, perhaps two dives. This time estimation includes both the level-1 and level-2 protocols. If you can include more people, it should go faster but you will have to train them to reduce variability in data collection and provide the materials.

4. Data for size classes will be put in a separate spreadsheet with date, locality, site depth interval, and urchin size.

MATERIALS NEEDED

Plastic transect tape
chain
re-bars (optional)
UW slate with glued section of measuring tape
Large plastic basket
Long BB-Q thongs and fork
Two pointed compass
Random number table.





Data information should be sent to the project leader as soon as you have it. Everybody is welcome to participate. A manuscript will be put together for publication next year.

CORAL-OCTOCORAL DISEASE SURVEY - PROTOCOL LEVEL-2 CARICOMP

Modified by E.Weil
June of 1999

OBJECTIVES:

To assess the occurrence (incidence at population and community levels) and local/geographic distribution of coral and octocoral diseases across the wider Caribbean region.

More specifically : 1- To quantify relative frequencies of infected colonies in each coral species affected within and across localities. 2- To quantify relative frequencies of the different diseases within and across species and within and across localities. 3- To determine if there is a correlation between frequencies of disease corals with depth, and distance to anthropogenic affected areas.

LEVEL- 1 - PROTOCOL:

Protocol will work with many different sampling methods (quadrats, chain transects, belt transects, etc.). This is to be carried out at the two reef sites of the CARICOMP reef monitoring using the chain. The survey is organized in a hierarchical way that will facilitate the decision-making process. A set of bilingual-underwater-laminated cards (Bruckner & Bruckner & Weil) was selected to be distributed amongst all site directors. This ID cards are to be used as a the ONLY guide to identify the different diseases and other causes of coral mortality. This will serve as a tool to standardize the identification of the different diseases and causes of mortality. There are ten, 10 m long chain sections per locality. Survey should be conducted over a belt transect of two meters wide (one meter on each side of the chain) by ten meter long (20 m^2), for a total of 200 m^2 per locality at one depth interval. All coral and octocoral colonies within this area should be checked and counted. The sample unit is one colony (ramet). Each coral colony should be identified to species (or genus at least) and/or to either one of the five colony form descriptors specified in the CARICOMP manual (branching, crustose, massive, foliose or milleporid). Octocorals should be ID at least at the genera level, or using the four cathegories (Ros, fan, feather, whip) in the manual.

The first discrimination-category for each colony is if it is healthy or unhealthy. Healthy colonies are those without signs of disease, recent injury and/or bleaching. Unhealthy colonies are those with any signs of disease, recent, open injuries and/or bleaching. A non-healthy colony will then be put into one of three categories, bleached, injured or diseased. If a colony has a disease and any of the other two signs (bleaching, injuries) it will be included in the disease category. Notes about the other symptoms taken. Bleaching should be verified by looking for the presence of live tissue (but decolored) and at the pattern of bleaching (usually not a band-like pattern like most “white-type” diseases; e.g. **pale, blotchy or white**). Injured colonies include those with damage from predation (often with skeletal damage), anchors or other physical damage, and overgrowth. If you can identify the type of damage (small lesions like individual bite marks or large excavations such a anchor damage) please do so. If the colony is being overgrown by other organisms (algae, zoanthid, sponge, hydroid, tunicates, etc.) it is important to note which organism (s) are overgrowing it. Colonies with old dead areas covered with algae but with healthy looking tissues fall into the healthy category.

A diseased colony will then be carefully observed to identify the disease that is affecting it. Observations should be compared with the information in the Q-cards to put the colony within any of the

following 9 categories (see below and the flow diagram):

1- Black-Band Disease (BBD)	Multispecific
2- White-Band Disease (WBD)	Types I and II (only in acroporids for the moment)
3- White Plague-II (WP-II)	Multispecific
4- Yellow-Blotch Disease (YBD)	Only described in species **of** *Montastraea* but also reported in many other species.
5- Dark Spots Disease I (DS-I)	Small, dark areas with no aparent tissue mortality. The one common in *Siderastrea spp*.
6- Dark Spots Disease II (DS-II)	Large dark areas, larger than the DSD-I that is common in *M. annularis* and *S. intersepta*.
7- Red Band Disease (RBD)	**Careful** here since BBD can be seen as red bands too. RBD has been reported for *Gorgonia* spp and Agaricids in the Caribbean.
8- Aspergillosis (ASP)	In *Gorgonia ventalina, G. flabellum, P. americana* and other octocoral species as well (*Plexaura flexuosa*).
9- Other -	this category includes all other "unconfirmed-pathogen-produced" diseases (tumors, hyperplasia, white pox, and all the "blotches").

COLONY

Identify colony to species or any of the other categories, (branching, massive, foliose, etc). Is the colony?

HEALTHY | **UNHEALTHY**

BLEACHING
Check for presence of tissue and pattern of bleaching)

DISEASED

INJURED
(Damaged by : Predation, anchors overgrowth, etc)

IDENTIFY DISEASE

BBD | **WBD** | **WP-II** | **YBD** | **DS-I** | **DS-II** | **RBD** | **ASP** | **OTHER**

LEVEL 2 PROTOCOL - CORAL DISEASES

The purpose of this activity is to assess the incidence of diseases in each coral and octocoral species at different depth intervals (habitats) and in at least two different reef sites per locality. The diseases will be identified following the protocol level-1 described above. The field experimental design is as follows :

1. At least two widely separated (more than 5 km) reef sites should be selected at each geographic locality (CARICOMP chapter), and preferably, one near and one far from anthropogenic-impacted areas. A preliminary qualitative, visual assessment of current levels of disease incidence and depth distribution within the two different reef localities should be done to determine if coral diseases are common or rare.

2. A quantitative assessment will follow using a modified protocol from the standard CARICOMP coral survey. Three depth intervals would be selected in those reefs with a depth profile to at least 45 feet deep. The depth intervals are: 0-15 feet; 16-35 feet and 35-50 feet, so major habitats would be surveyed (shallow, branching *Acropora* zone and boulder-crustose platform zone; intermediate depth *Montastraea* zone - massive, boulders and platy spp.; and the deeper more diverse zone - foliose, massive and platy spp). Re-bars or other permanent means of marking the transect position should be hammered into the substrate for future monitoring.

In each depth interval, at least three 20 meter long x 2 m (40 m²) wide band-transects should be surveyed (a minimum of 9 transects/reef). Two sets of 6 band-transect each should be separated at least 100 m (see diagram below). In each depth interval a set of two band-transects could be done using a 50 m long, plastic tape. Once the tape is positioned, two 20 x 2 m bands separated by a 10 m interval will be surveyed (from 0 to 20 m and 30 to 50 m). A one m long pvc section, marked in cm (100 cm) will be moved along each side of the transect line and every colony of coral and of *Gorgonia* spp. will be surveyed using the protocol level 1 described above. Once a colony is found to have a disease ,two diameters (maximum and minimum) will be measured with the pvc stick to estimate surface area. **ONLY disease colonies (corals and Gorgonia spp) will be measured**,

Separation between the consecutive band transects (at same depth interval) should be at least 10 m. If the reef areas do not have a depth profile or slope, or are such that only two depth intervals can be selected, the minimum of 12 band-transects should be then spatially distributed within the area. All coral colonies should be recorded to species (or genus at least) level.

Confusion of what an individual colony or ramet is will probably arise when branching, columnar or massive species that have suffered fission (fragmentation and/or partial mortality) are encountered. For the purposes of this protocol, a ramet is any colony that is spatially separated from any other colonies of the same species. On some occasions however, it is clear for some fragmented colonies, that in the near past, there was a physiological and physical connection between two or more ramets (i.e. in massive colonies suffering partial mortality or broken branches nearby the mother ramet of ramose species). In these situations, instead of counting 2000 small fragments (i.e. *P. porites* or *M. mirabilis*) one should use common sense and spatially delimit the spread of the individual genet (in the many ramets) and count it as a single ramet (genet).

The study is aimed at comparing the incidence of coral diseases between different reefs within a given location (Cuba, Panama, Venezuela, etc.) and across locations in the wider Caribbean. It is fundamental to the proposed experimental design to insure the sampling of large reef areas, in order to estimate current levels of disease incidence at a suitable spatial scale. Time window to conduct the surveys is

between July and October. Frequency of surveys will depend on each individual laboratory's logistics, time and personnel.

A template to input data is available at the CARICOMP website (in Microsoft Excel format).

ALGAL BIOMASS SURVEY

Equipment:

Suction dredge
SCUBA tank
20 cm metal ring
Spatula
Salad spinner
1-2 mm mesh bags

Procedure:

To determine algal biomass use a suction dredge fitted to a compressed air tank to collect algae in a nylon bag with a 1-2 mm mesh (Figure 1). Collect the algae from inside a ring 20 cm in diameter in which the algae were scraped with a spatula, while suction was being applied. At least 5 (preferably 10) random samples must be taken from each station, care being taken to avoid the inclusion of corals, sponges or large gorgonians (> 5 cm).

The algae must be washed with seawater to remove sediments. Excess water is drained by placing the algae in a salad spinner, and spinning for 30 seconds. The algae are then weighed. The biomass is expressed as grams wet weight per square meter.

All species found at each station must be recorded and the five most abundant species weighed separately after combining all the samples from each station.

Sampling should be carried out at least twice a year, between March and April, and between September and October.

Data that must be included:

Site
Station
Date
Wet weight
Biomass as g per square meter
All species of algae
The 5 dominant species
Dominant species biomass