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A PHOTOGRAPHIC ATLAS OF
NORTH FLORIDA ESTUARINE
PHYTOPLANKTON
AND A SUMMARIZATION OF LIFE HISTORY
RELATIONSHIPS AND ASSOCIATIONS WITH
VARIOUS ENVIRONMENTAL CONDITIONS

by

Robert J. Livingston and A. K. S. K. Prasad

**Center for Aquatic Research and Resource Management
Florida State University
Tallahassee, Florida 32306**

December, 1989

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PREFACE

This report is based on years of studies in the Choctawhatchee River and Bay system and the offshore Gulf of Mexico. The data are currently undergoing analysis, modelling, and publication in a series of reviewed scientific publications. This data base is part of a more extensive research effort by the Center for Aquatic Research and Resource Management.

ACKNOWLEDGEMENTS

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Funds for this project were provided by the Department of Environmental Regulation, Office of Coastal Management using funds made available through the National Oceanic and Atmospheric Administration under the Coastal Zone Management Act of 1972, as amended. Mr. James W. Stoutamire administered the grant on behalf of the FDER.

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LIST OF FIGURES

- Figure 1: Maps of the study area for the phytoplankton collections including the Choctawhatchee River and Bay systems and associated Gulf areas (Station-specific data listed in Appendices I and II)
- Figure 2: Summary data (12 month averages) of physical-chemical factors taken in the Choctawhatchee Bay system from September, 1985 through August, 1986.
- Figure 3: Summary data (12 month averages) of chlorophyll a taken in the Choctawhatchee Bay system from September, 1985 through August, 1986.
- Figure 4: Summary data (12 month averages) of particulate organic matter taken in the Choctawhatchee Bay system from September, 1985 through August, 1986.
- Figure 5: Summary data (12 month averages) of phytoplankton numbers and species richness taken during the day in the Choctawhatchee Bay system from September, 1985 through August, 1986.
- Figure 6: Summary data (12 month averages) of physical-chemical taken in the Choctawhatchee Bay system from September, 1985 through August, 1986.
- Figure 7: Phytoplankton data (64 μ m nets) concerning numbers (X 1000) per m³, species richness, and Shannon-Wiener diversity taken monthly from September, 1985 through August, 1986.
- Figure 8: Phytoplankton occurrence as a function of distributions of salinity, dissolved oxygen, Secchi depths, orthophosphate, nitrate, ammonia, and chlorophyll a. The numerically dominant phytoplankton species (16) are tested as possible indicators of water quality phenomena in the Choctawhatchee Bay system from September, 1985 through August, 1986.

LIST OF TABLES

- Table 1: Systematic review of phytoplankton taken with 25 μm nets in the Choctawhatchee River system.
- Table 2: Cluster analysis (Czekanowski similarity coefficient; flexible grouping strategy with $\beta = -0.25$) of stations by sampling parameters (Choctawhatchee estuary, all dates, surface and bottom station data pooled).
- Table 3: Metal accumulation index (A) computed for seven metals at 25 locations sampled in sediments of Choctawhatchee Bay in April, 1987.
- Table 4: Systematic review of phytoplankton taken with 25 and 64 μm nets in the Choctawhatchee estuary from September, 1985 through August, 1986.

EXECUTIVE SUMMARY

An analysis was made concerning the phytoplankton taken in the Choctawhatchee River and Bay system and offshore portions of the Gulf of Mexico. A photographic atlas of the algae taken in the Choctawhatchee Bay system was featured along with a determination of how well such organisms were indicators of water quality in this estuary. Various community parameters such as numbers per unit volume, species richness, and species diversity were useful in identifying specific water quality conditions associated with cultural eutrophication in the bay system. Some dominant species were identified with specific water quality factors although the use of single species indicators of such water quality is complicated by the fact that many estuarine species have broad tolerances to such conditions. A combination of single species and community factors may be useful as indicators of various forms of aquatic habitats including water that is affected by anthropogenous activities. Data analyses will continue with an emphasis on combinations of moderately abundant and rare species as possible indicators of water quality from fresh water to estuarine and marine systems.

TABLE OF CONTENTS

	Page
Preface	i
Acknowledgements	ii
List of figures	iv
List of tables	v
Executive summary	vi
I. Introduction.....	1
A. Qualitative algal indicators	
B. Choctawhatchee basin characteristics	
C. Geomorphology	
D. Climate	
E. Land use and water quality	
II. Methods and materials.....	12
A. Water quality	
B. Phytoplankton	
III. Photographic atlas.....	18
IV. Analysis of species distribution.....	19
A. River algae	
B. Estuarine phytoplankton distribution	
1. Water quality	
2. Taxonomic review: species occurrence	
3. Algae as water quality indicators	
V. References.....	31
VI. Appendices.....	35

I. INTRODUCTION

The use of algae as indicators of water quality is well developed in the scientific literature (Schubert, 1984) and has been developed along both qualitative and quantitative lines. The background of such methods in the field includes a general acceptance that clean water supports speciose assemblages of algae whereas polluted water tends to reduce the number of species and increases the relative dominance due to the survival and well-being of a few resistant forms. There is, however, relatively little information concerning the microalgae of the major drainages in Florida, especially along the northern gulf coast.

A. Qualitative algal indicators

Phytoplankton are microscopic aquatic plants that are usually free-floating and suspended in a series of habitats, "having little or no resistance to currents" (APHA/AWWA/WPCF, 1980). The use of such organisms as indicators of water quality is well established. This includes the use of various species that either flourish in highly eutrophic conditions and/or are sensitive to various types of toxic wastes. According to such information, there are certain algal indicators that can be used in the evaluation of water quality. Clean water indicators include *Melosira islandica*, *Cyclotella ocellata*, and *Dinobryon* spp. Indicators of contaminated water include *Aphanizomenon flos-aquae*, *Microcystis aeruginosa*, and *Nitzschia palea*. Some forms, such as the former two species, can be associated with algal blooms and anoxic/hypoxic conditions. According to APHA/AWWA/WPCF, 1980, the following groups can be used as indicators:

Clean water algae

Agmenellum
Ankistrodesmus
Calothrix
Chromulina
Chrysococcus
Cladophora
Coccochloris
Cocconeis
Cyclotella
Entophysalis
Hildenbrandia
Lemanea
Meridion
Micrasterias
Microcoleus
Navicula
Pinnularia
Rhizoclonium
Rhodomonas
Staurastrum
Surirella
Ulothrix

Pollution indicators

Anabaena
Arthrospira
Carteria
Chlamydomonas
Chlorella
Chlorococcum
Chlorogonium
Euglena
Gomphonema
Lyngbya
Nitzschia
Oscillatoria
Phacus

Phormidium
Pyrobotrys
Spirogyra
Stigeoclonium
Tetraedron

There are problems with the use of such indicators. Because of the temporal and spatial variability, the definition of water quality by microalgal species can be difficult. In rivers, the origin and level of exposure of algae to various water types remains problematical. In estuaries and marine systems, complex currents also can preclude exact definitions based on the presence and/or absence of algal indicators. Presence/absence data are qualitative; in this way, the use of indicator species is limited by the problems associated with qualitative methods of assessment. Such assessments cannot lead to the evaluation of cause-and-effect relationships. In addition, process oriented functions cannot be determined using indicator species.

On the other hand, the use of algae as indicators of water quality has a well established basis (Stoermer, 1984). Qualitative data can be used to determine the species associations of a given aquatic system. The use of correct systematic data is crucial to the determination of the ecosystem structure. Such information is important in the interpretation of process-oriented data. For instance, due to gas vacuole formation, an ability to escape nitrogen limitation, and a colonial growth habit, blue-green algal species of the genera *Anabaena*, *Anacystis*, and *Aphanizomenon* are considered indicative of water pollution of various types. Some forms of blue-green algae also emit toxins that can have an adverse effect on other species. Thus, the blue-green algae are important in the evaluation of water quality. In marine systems, dinoflagellates have been implicated in the release of toxins. Flagellates can cause problems in brackish water ponds. The species *Oscillatoria rubescens* is an indicator of the eutrophication of oligotrophic lakes (Edmondson, 1972). There is a succession of algal species as eutrophication proceeds. According to Reddy and Venkateswarlu (1986), pulp mill

effluents caused certain changes in riverine algal assemblages. There were reduced numbers of phytoplankton in the effluent channel; The Cyanophycean types were dominant in such areas with an almost total lack of green algae. *Oscillatoria* spp were the dominant species in the polluted areas with *Rhopalodia gibberula* and *Nitzschia palea* as the primary diatom species. Premila and Rao (1977) showed that blue-green algae (*Oscillatoria nigroviridis*) were indicative of waters characterized by sewage relative to less polluted areas. Low salinity in such areas tended to encourage this species along with the sewage effluents. Blue-green algae are also abundant in marine areas under natural conditions (Potts, 1980). Qualitative signs can thus be used in the identification of water quality problems.

Diatoms are useful in the analysis of the history of a given water body (Stoermer, 1984). According to Schoeman and Haworth (1984), diatoms can be very useful under certain circumstances in the evaluation of water quality. Such algae are: easy to collect; cosmopolitan; responsive to environmental conditions (short and long-term); relatively well understood ecologically; suitable for historic surveys; suitable for diversity analyses. Disadvantages include the fact that identification is highly technical and requires considerable expertise. Patrick (1984) has reviewed the manner in which shifts of diatom assemblages can be used to indicate changes in ambient water quality conditions. Different species of diatoms flourish under varying concentrations of nutrients and pollutants. Often, the response of a given population may be indirect, due to the effects of increased or decreased competition from species that are affected by water quality conditions. Geissler and Jahn (1984) have reviewed the infraspecific taxa with respect to morphological and ecological differentiation. Sullivan (1984) showed that diatoms can be quantitatively used in freshwater systems but that the use of such populations in estuarine and marine systems is much less well developed and understood. Salinity changes, together with highly variable (spatial and temporal) distributions of diatoms, have not been well studied. Thus, the actual relationships of diatoms to water quality in estuarine and marine systems are not as well developed as

they are in freshwater areas. Wilderman (1984) has outlined the indicator diatoms for spatial and seasonal variation. Although there are identifiable salinity and temperature preferences of estuarine diatoms and that seasonal changes in such indicators are relatively easy to establish, the distribution patterns within seasons are more difficult to interpret. Euryhaline species complicate the process in that they are not useful indicators of salinity distribution. The complexity of the diatom assemblages in estuarine areas was illustrated by Squires and Sinnu (1982) who showed that various factors are related to the determination of diatom distribution. Currents and salinity were important factors in the determination of diatom assemblages in the area of study. Marshall (1982) showed that the marine diatom distributions in the Gulf of Maine were related to the location of large bay systems and the Georges Bank.

Maestrini et al. (1984) gave a complete review of the use of algae as indicators of water quality in marine systems. The authors point out the various problems of trying to make associations of water quality factors (and nutrient processes) and descriptive algal data. Differences in essential nutrients, the physiological state of the indigenous algal populations, and other factors that are difficult to measure all contribute to the complexity of causal relationships in estuarine and marine systems. The authors advocated the experimental (e.g., bioassay) approach to evaluating water quality relationships with phytoplankton. Actually, both methods (descriptive and experimental) are necessary for a real understanding of how aquatic systems function. In any case, an evaluation of the species composition of the algae in a given system represents an important first step in the evaluation of existing water quality.

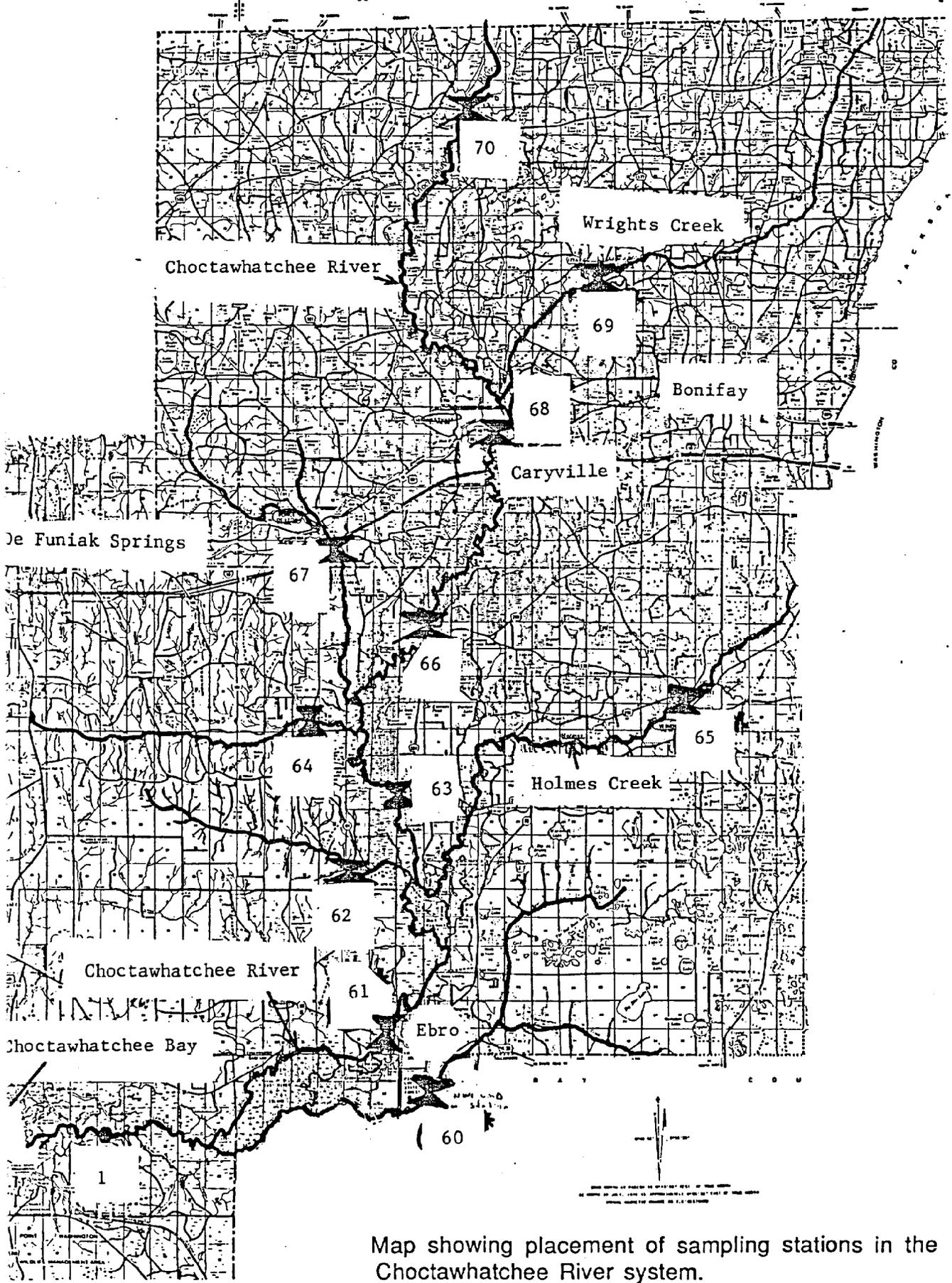
The toxic effects of contaminants on algal communities in the field have been addressed only recently. This entire subject is, in fact, relatively complex and poorly understood. At the ecosystem level, the natural interactions (physical and biological) of the phytoplankton in freshwater and marine systems along the northern

Gulf coast of Florida remain virtually unknown. Unless ambient field conditions are established, the influence of human activities on algal associations in a range of habitats will be difficult to evaluate. This atlas is an attempt to establish the ambient conditions of phytoplankton assemblages in a series of aquatic habitats (freshwater, estuarine, marine) in a relatively unpolluted (the Choctawhatchee River-bay) system with some attention to the associated offshore (Gulf) region.

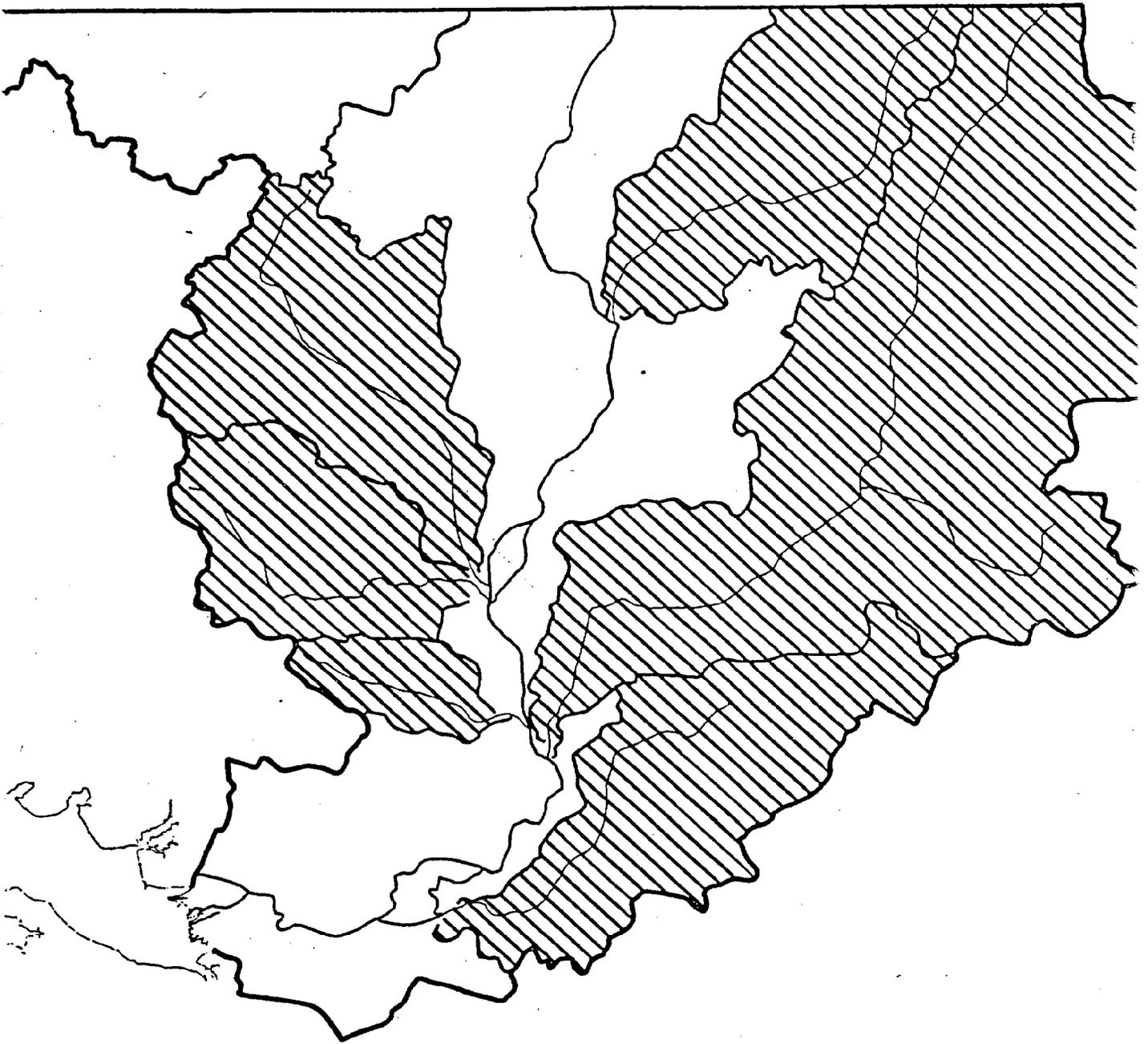
B. Choctawhatchee Basin characteristics

Detailed reviews of the basic geomorphology and hydrology of the Choctawhatchee River basin are available (Abbot, Merkt & Company, 1960; U. S. Study Commission, Southeast River Basins, 1963; Federal Power Commission, 1966; Soil Conservation Service, U. S. Department of Agriculture, 1975; Northwest Florida Water Management District, 1980, 1988; U. S. Army Corps of Engineers, 1980).

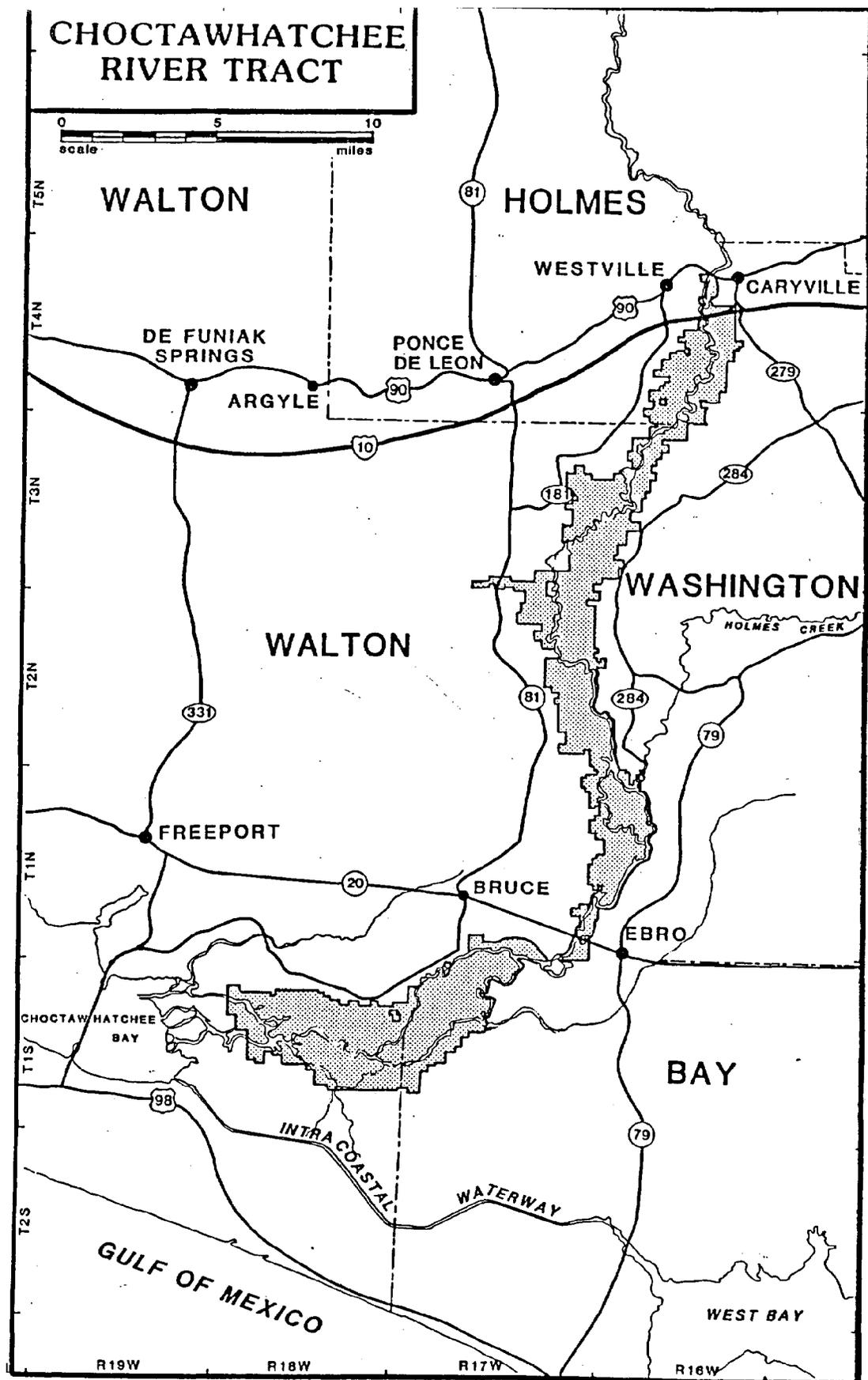
The Choctawhatchee River system represents the main source of fresh water for the Choctawhatchee Bay system, and is the fourth largest river in terms of flow volume in the state of Florida. The Choctawhatchee River is formed by a series of tributaries in Alabama and Florida (Figure 1). The headwaters are in Barbour County, Alabama. The major tributary, the Pea River, joins Whitewater Creek to form the western wing of the Choctawhatchee drainage basin as it joins the main river just south of Geneva, Alabama. In Alabama, the Choctawhatchee River runs for about 88 miles draining approximately 2,500 square miles. In Florida, the river then runs another 87 miles to drain about 1,500 square miles. Here, the major tributary is Holmes Creek. Other major tributaries to the Florida portion of the river include Wright's



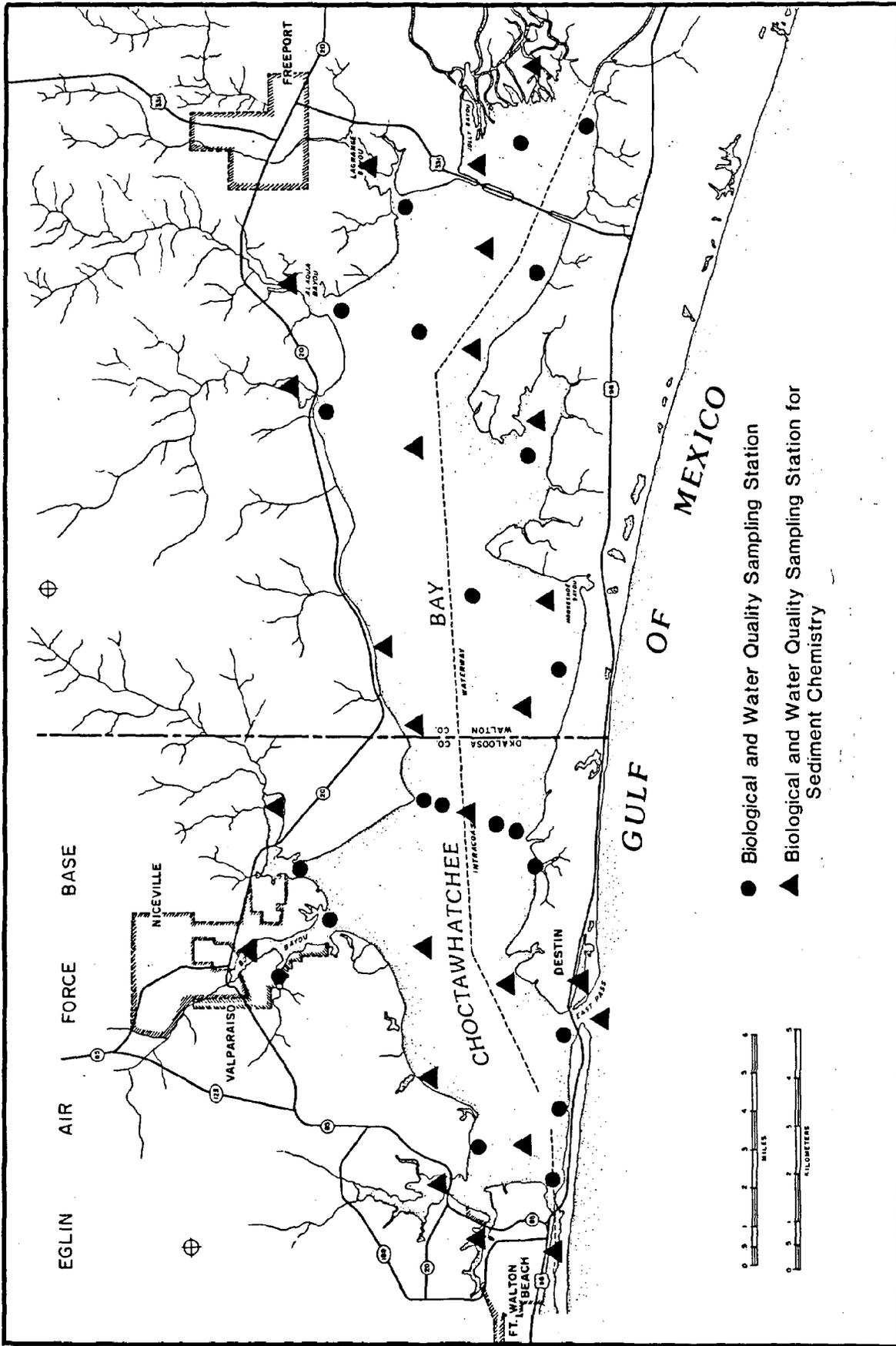
Map showing placement of sampling stations in the Choctawhatchee River system.



Map showing the various sub-basins within the lower Choctawhatchee River system (courtesy of the Northwest Florida Water Management District, Mr. Tom Pratt).



Map showing extent of the recently purchased wetlands (state purchase of the Choctawhatchee River Tract; Northwest Florida Water Management District) within the area of study. (courtesy of the Northwest Florida Water Management District)



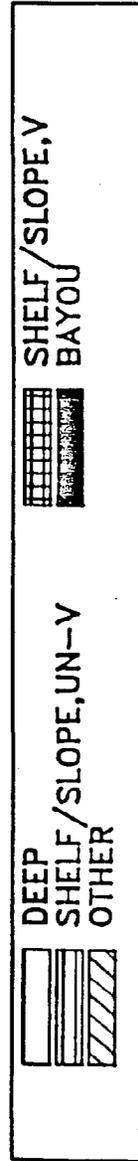
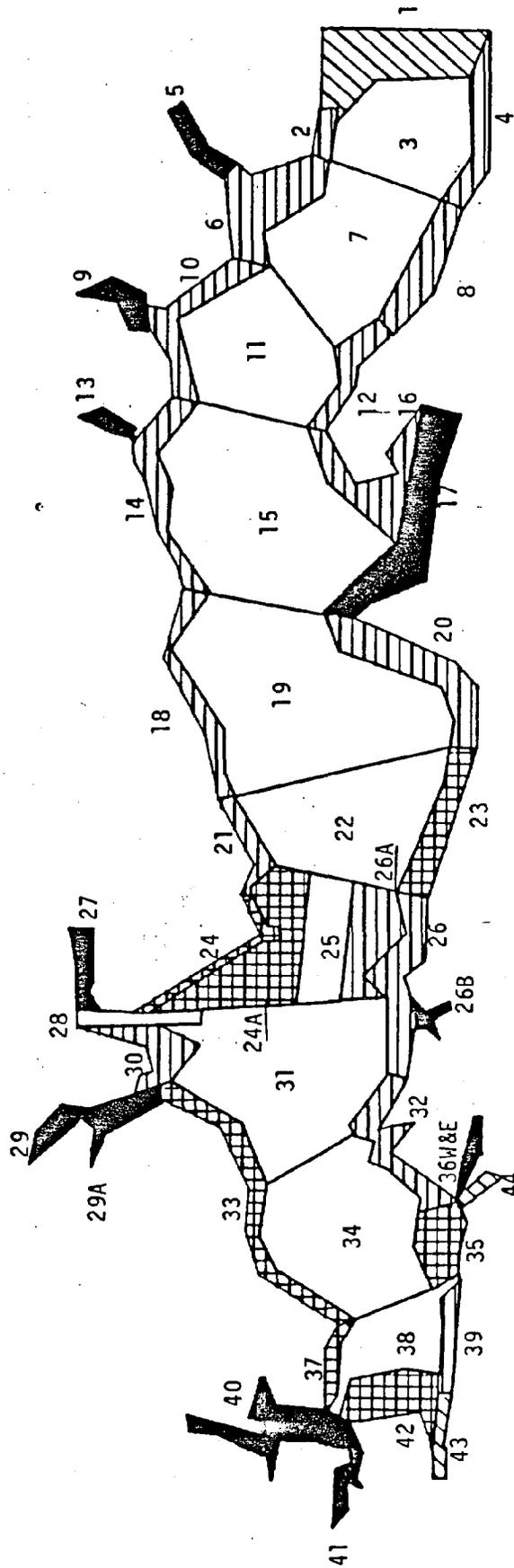
Location of Sampling Stations

Computerized map of habitat distribution by station in the Choctawhatchee Bay system.

CHOCTAWHATCHEE BAY

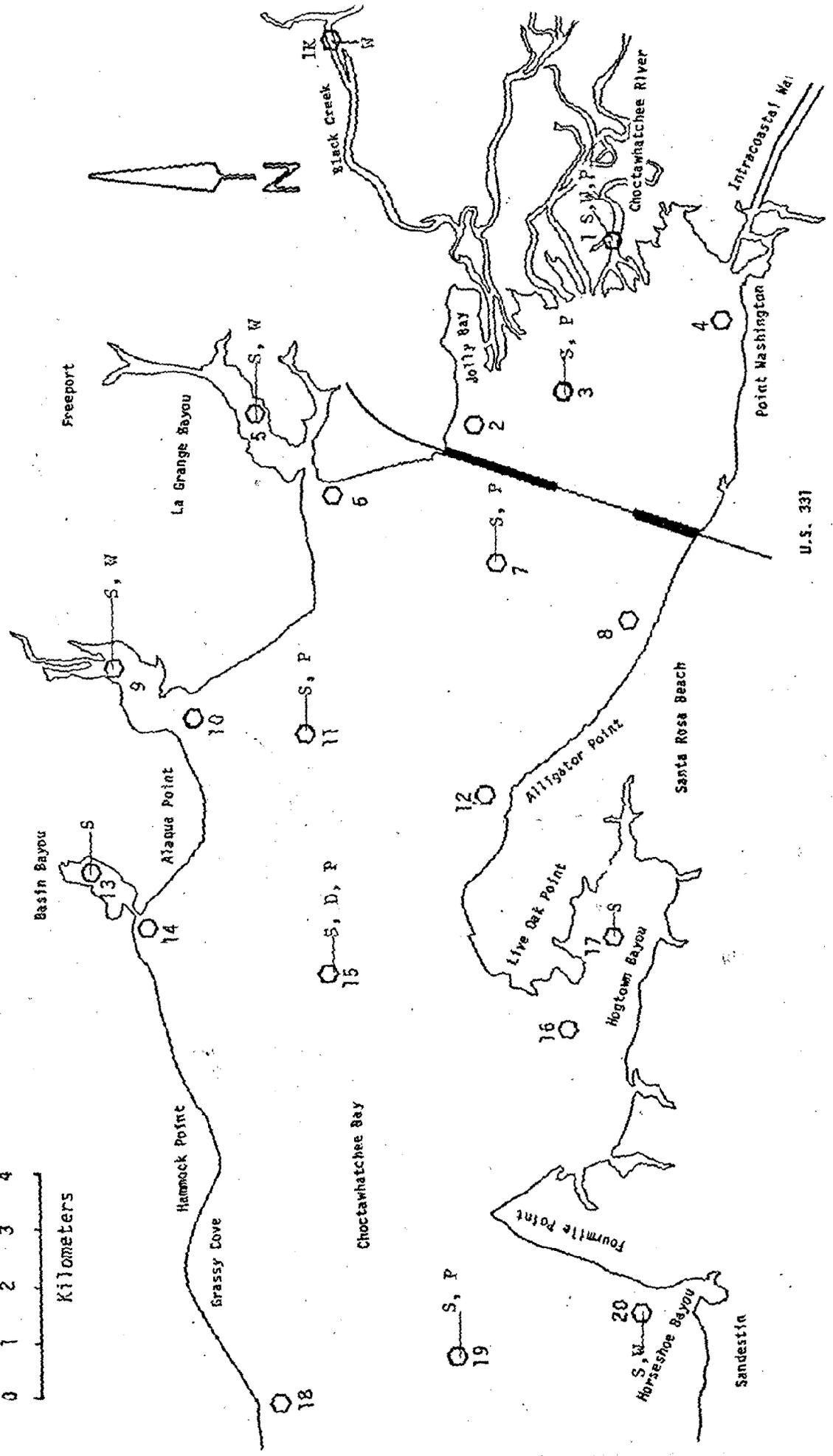
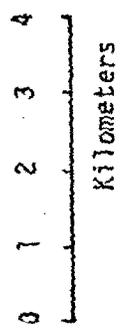
Habitat Distribution

BY STATION



Place names/approximate station locations:
 Choctawhatchee Bay Project 1985-1986

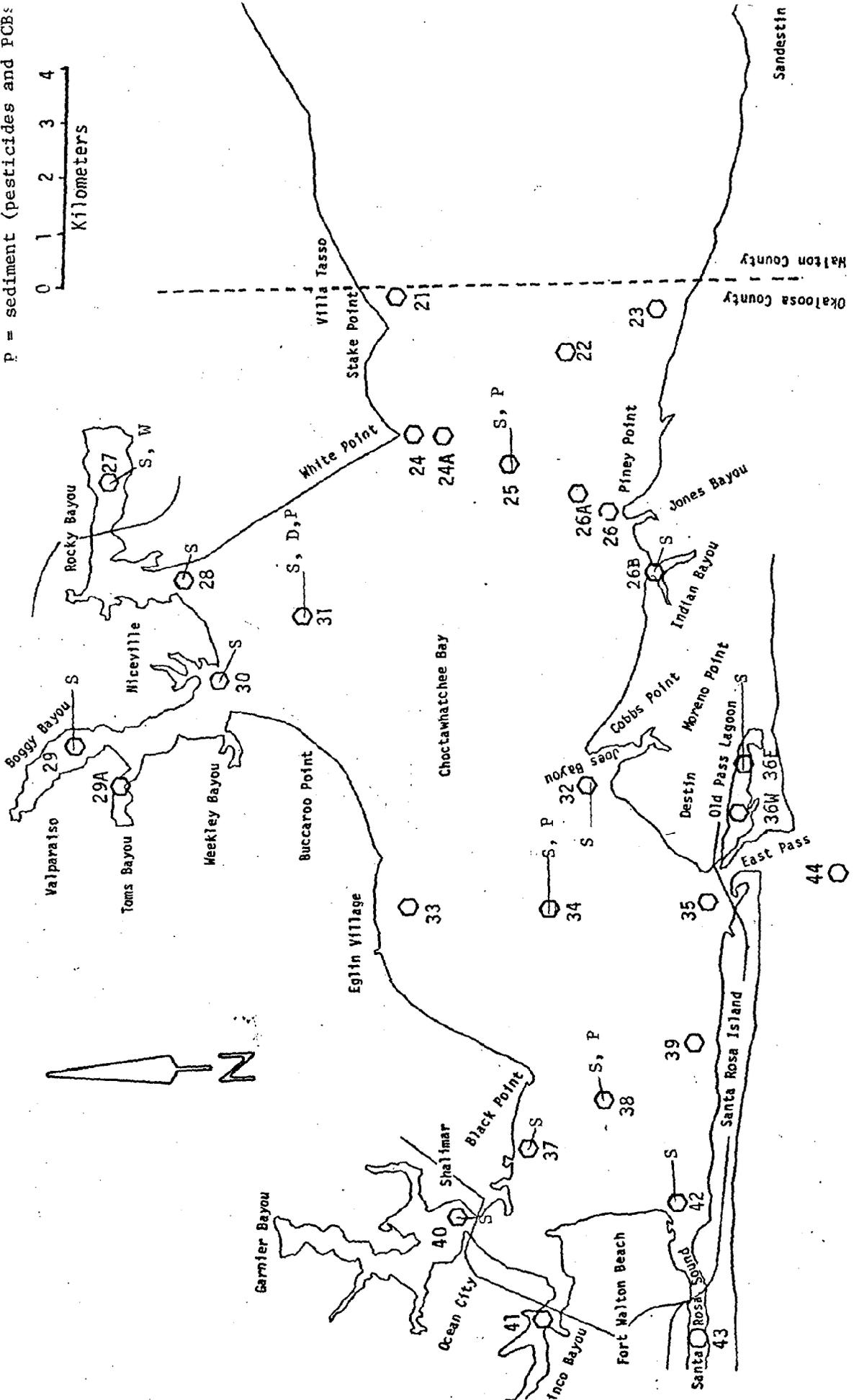
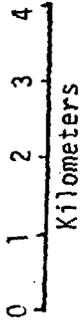
S = sediment sample (me-)
 W = water sample
 D = sediment sample (dioxin only)
 P = sediment (pesticides and PCBs)



U.S. 331

Place names/approximate station locations:
 Choctawhatchee Bay Project 1985-1986

S = sediment sample (metals)
 W = water sample
 D = sediment sample (dioxin only)
 P = sediment (pesticides and PCBs)



CAPE ST. GEORGE TO MISSISSIPPI

R.
Radar reflector
floating aid
reflector ident
omitted from 1

Mercator Projection
Scale 1:456,394 at Lat. 29°
North American 1927 Datum

SOUNDINGS IN FATHOMS AT MEAN LOWER LOW WATER

(For offshore navigation only)

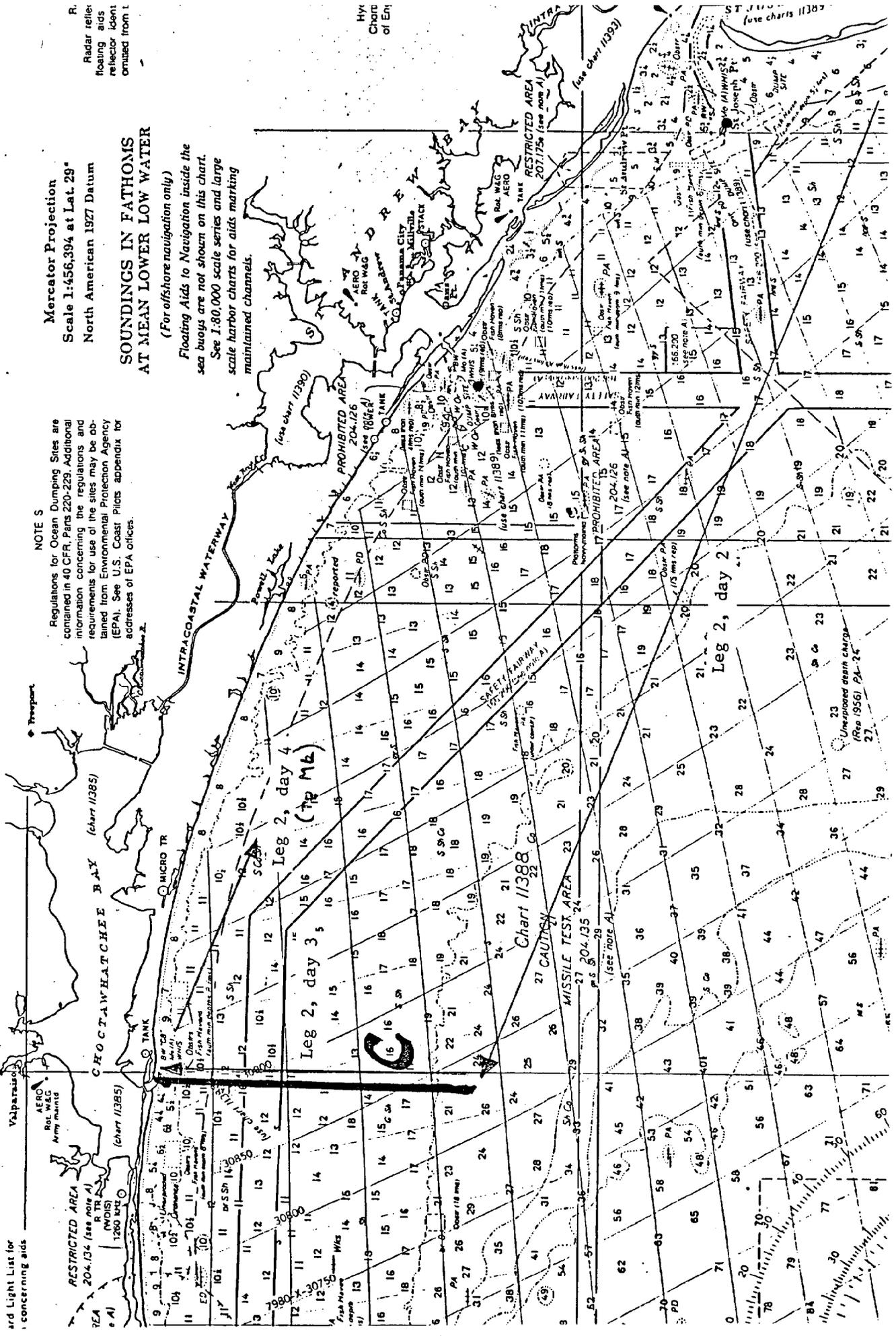
Floating Aids to Navigation inside the sea buoys are not shown on this chart. See 1:80,000 scale series and large scale harbor charts for aids marking maintained channels.

NOTE S

Regulations for Ocean Dumping Sites are contained in 40 CFR, Parts 220-229. Additional information concerning the regulations and requirements for use of the sites may be obtained from Environmental Protection Agency (EPA). See U.S. Coast Pilots appendix for addresses of EPA offices.

Fish Haven in the Safety Fairway area is 47 feet.

members shown with
STATION
and Light List for
concerning aids



Hyd
Chart
of En

Leg 2, day 3
Leg 2, day 4 (TP Mb)

Chart 11388
22 CAUTION

MISSILE TEST AREA
23 PA 25
(see note A1)

Leg 2, day 2

Unreported depth changes
(Reo 1956) PA-25

Creek, Sandy Creek, Bruce Creek, Seven Run Creek, and Pine Log Creek. Sampling stations were placed on each of these tributaries in addition to main stem (Figure 2). Such tributaries represent a major part of the basin for the Florida portion of the river (Figure 3). These stations also include major sections of the wetlands recently purchased by the state of Florida (through negotiations by the Florida Northwest Water Management District) as shown in Figure 4. The average flow rate of the river ranges between 5,500 and 7,000 cfs (U.S.G.S., 1978) with low flows approximating 3,400 cfs and high flows of 26,000 cfs (Ross et al., 1974).

The Choctawhatchee Bay system (reviewed by Livingston, 1986a) is an east-west oriented estuary with the primary freshwater input at the east end via the Choctawhatchee River (Figure 1). Fresh water also enters the system along a series of smaller drainage areas primarily in northern sections of the bay. River flow peaks occur during winter-spring months with low flows in the summer and fall. There is a shallow shelf that is located around the fringes of the bay; sandy sediments occur here. The central portions of the bay are deeper and are characterized by fine-grained, highly organic sediments. The spring temperature transition occurs in March with peaks of temperature from June to August. The fall transition occurs in November with winter lows from December to February. Salinity gradients follow trends in river flow with the lowest salinities found from December through April. Pronounced vertical salinity stratification occurs in large portions of the bay (particularly in western and central areas); during warm periods, such stratification is associated with hypoxic conditions at depth. In August, 1986, virtually the entire bay was hypoxic to anoxic at depth. Such hypoxia occurs in various western bayous and lagoons (lower Rocky, Boggy, Tom's, Garnier, Cinco, Old Pass Lagoon). Such areas are adversely affected by storm water runoff and other factors such as

marinas (Old Pass Lagoon). Nitrogen levels are highest in western sections of the bay and phosphorous is highest in Old Pass Lagoon, Lower Rocky Bayou, and Boggy Bayou.

C. Geomorphology

Surface sediments, composed of lime accumulations and/or sedimentary deposits of sand, silt, or clay, rest on a base of crystalline rock some 2,500 to 4,000 feet below (U. S. Study Commission, 1963). The lower coastal plain is flat and sandy with beach ridges extending to elevations of up to 200 feet. The Tertiary limestones, forming the principal artesian aquifer in this part of Florida, form outcrops at or close to the surface of the bed of the Choctawhatchee River. The Ocala limestone, with its sinkhole topography due to the solution of the limestone bedrock, forms a major portion of the Deadening Lakes region of the Choctawhatchee drainage in Washington County, Florida at the southern end of the river.

D. Climate

The Choctawhatchee River basin lies in a south temperature region characterized by mild winters and hot/humid summers. Some moderation of these climatological characteristics occur in the coastal region of the basin. The average temperature is 68 °F with a range of 50 °F in December to 81 °F during July/August (U. S. Study Commission, 1963). Average annual rainfall varies from 52 inches in the upper basin to 62 inches in the southwest portion of the system. Maximum average annual precipitation approximates 85 inches (1929) with the minimum such levels about 26 inches (1954). The wettest months are

June-September. The Floridan Aquifer provides the major source of water to the river system which includes numerous streams, lakes, and springs. The maximum 24-hour precipitation was recorded as 20 inches at Elba, Alabama in March, 1929 (U. S. Study Commission, 1963). During the spring of 1975, there was heavy rainfall and flooding (17.7 inches in a 24 hour period in April, 1975); this stimulated the evaluation of storm control measures (U. S. Department of Agriculture, 1975) due to what were perceived as major flood and erosion damage. Various flood control measures have been proposed by the U. S. Army Corps of Engineers for the Choctawhatchee basin (Northwest Florida Water Management District, 1980). Such proposals include structural and non-structural methods along with land use regulations.

E. Land use and water quality

Land use within the Alabama portion of the Choctawhatchee basin is dominated by forestry (51.7%) and agriculture (cropland, 30.6%; pasture, 11.6%) (Alabama Water Improvement Commission, 1976). There is relatively little urban development in the region (3.2%). In Florida, the basin remains largely undeveloped with forestry (58.4%) and agriculture (25.7%) as the major land uses (Florida Department of Environmental Regulation, 1980). The largest cities in the Choctawhatchee basin include Chipley, Bonifay, and Defuniak Springs. The western section of the bay is becoming highly urbanized with stormwater runoff bringing nutrients and various pollutants into the system. Such urbanization is still not present in eastern sections of the bay so an east-west gradient of water quality exists in addition to the above-noted salinity gradient.

The major sources of pollution are agricultural runoff, sewage discharges, and minor industrial effluents in the river basin and stormwater runoff and marina wastes in the bay system. According to the Florida Department of Environmental Regulation (1980), the mean dissolved oxygen levels in the river were above the state water quality criterion (5.0 mg/L). The pH levels in the river varied from 6.3-7.6. There were 'low' levels of Kjeldahl nitrogen (mean X = 0.03-0.05 mg/L), nitrate/nitrite (mean X = 0.06-.26 mg/L), and total phosphorous (mean X = 0.03-0.05 mg/L) throughout the river with a decreasing trend toward the mouth of the river. Concentrations of mercury, cadmium, and lead were in excess of the state water quality criteria. High levels of fecal coliform bacteria were noted near the town of Ebro due to what is thought to be agricultural runoff (Florida Department of Environmental Regulation, 1980). Trend analyses indicated increasing total phosphorous, decreasing nitrate-nitrite, increasing average dissolved oxygen, and decreasing mean pH. Available data indicated "generally good water quality in the Choctawhatchee River south of the Florida-Alabama State line" (Florida Department of Environmental Regulation, 1980).

More recent studies show significant water quality degradation in the lower Choctawhatchee basin. According to a recent update of this report (Florida Department of Environmental Regulation, 1986), the Choctawhatchee River basin now exhibits more water quality problem areas than other areas of low population density. There are high ambient values of nutrients in Wright's Creek near Noma, Florida due to agricultural runoff and possibly the numerous impoundments of this area. Upper Holmes Creek has water quality problems due to discharges from Graceville (Little Creek), Chipley (Alligator Creek), Vernon (Little Branch), and Bonifay (Camp Branch). Such problems are due

largely to sewage discharges. Holmes Creek also receives runoff from agricultural areas such as hog farms. West Sandy Creek also has degraded water quality due to sewage from Defuniak Springs and Bruce Creek receives effluent from a chicken processing plant. There were violations of standards concerning dissolved oxygen, NH_3 , coliform bacteria, and BOD_5 . Bioassays indicated toxic wastes. One area of Bruce Creek, near a 331 truck stop, has been polluted with oils and diesel fuel. The lower Choctawhatchee River was considered having "significant" biological degradation with a low number of macroinvertebrate species (Florida Department of Environmental Regulation, 1986).

II. METHODS AND MATERIALS

The general distribution of sampling sites is given in Figure 1. The field sampling program included different variables that were to be evaluated together to determine specific ecological features of the Choctawhatchee Drainage system. In the river, such variables included habitat characteristics, flow rates, water quality and sediment characteristics, mass flows of nutrients through the system, and biological features (e.g., phytoplankton, infaunal macroinvertebrates, epifaunal macroinvertebrates, fishes, food web organization). A sub-topic for analysis included the relationship between the various tributaries and the main stem of the river. The relationship of specific state variables such as river flow and wetlands distribution with the various biological features of the system were of major concern in this study. The sampling protocols for the estuary and offshore (Gulf) areas followed along similar lines (Appendix I).

A. Water quality

The methods used in the water quality analyses are detailed in Appendix II. Some additional nutrients (especially phosphorous compounds) were added to the original protocol.

River stations were sampled along with a series of established bay stations to evaluate the influence of the river on the bay. Routine water quality measurements included surface and bottom temperature, conductivity, dissolved oxygen, depth, and Secchi readings. In addition, three 2-l samples of

water were taken from the surface and bottom; these samples were iced and immediately shipped back to our Tallahassee laboratory for the analysis described in detail in our protocols.

B. Phytoplankton

Phytoplankton samples were taken with 15.2 cm (D) plankton nets (28 μ m mesh in the river and 64 μ m nets in the bay. In the river, the nets were suspended close to the surface on a line weighted at the bottom and attached to a float at the top. Three nets were set across the main stream for one hour. The center net had a flow meter suspended just below the net to quantify the volume of water sampled. At the end of the sampling hour, the nets were rinsed into numbered jars and preserved in 5% formalin (nets were numbered 1-3, which corresponds to left-right across the stream). In the bay, samples were taken with nets and with pumping as described in Appendix A. The eastern stations (3, 7,15) were sampled for two years with both 28 μ m and 64 μ m nets. The remaining phytoplankton stations (Appendix I) were sampled with 64 μ m nets.

The volume of each sample was measured in a graduated cylinder. The sample was then stirred with a magnetic stirrer for 1-2 minutes, and a 0.1 ml sub-sample was pipetted out. To limit clumping and cell damage, the stirrer was turned off between sub-samplings. Each sub-sample was placed in a Palmer-Mahoney counting chamber and the numbers of cells were counted by the 'strip' count method. In strip-counting, the top and bottom of the grid were the 'count' and 'no-count' boundaries, respectively, and plankters were counted as they moved across the center vertical line. Dead cells or diatoms with broken frustules were not counted. Empty centric and pennate diatoms were

counted separately as 'dead centric diatoms' or 'dead pennate diatoms' for use in converting the diatom species proportional count to a count per ml. A preliminary analysis (10 samples) was carried out to determine how many sub-samples were necessary for each count. Three subsamples provided a number within 22% of the mean of the 10 net sub-samples and this number was used for all counts.

All samples were processed to species. Diatoms were cleaned using heat if found by themselves. Otherwise, the sample containing various kinds of algae were placed in a 100 ml beaker and most of the water was decanted. A small amount of nitric acid was added and this solution was boiled for about 20-30 minutes. The sample was constantly stirred until oxidation of organic matter was complete. At this time, a small amount of potassium dichromate was added until the solution was brown in color. The solution was then cooled and decanted and distilled water was added. This procedure was repeated until the pH was 7. Alcohol was then added after the final washing and most of the water was decanted. Diatoms were mounted with Naphrax. A 0.01 ml aliquot of alcohol containing the cleaned diatoms was dropped on a coverglass so that the diatoms spread evenly; after the alcohol evaporated, the coverglass was heated so that the diatoms were incinerated onto the coverglass. The diatoms were then mounted in the Naphrax. After preparation, electron micrographs were taken on a Polaroid 4x5 Land film type 55/positive-negative using JEOL-JEM-100CX.II scanning and transmission electron microscope operating at an accelerating voltage of 20 KV. Light microscopy was also used with a Nikon biological microscope fitted with a phase condenser; photographs were taken on Kodak 35mm Panatomic-x fine grain black and white film using a Nikon camera. Phase contrast illumination was used for diatom studies. Soft-bodied

algae were photographed using wet mounts in distilled water. Methylene Blue and India ink were used to determine the extent of sheath formation. The structure of the pyrenoid and enclosed starch caps, flagellar number and insertion were studied with I₂ in KI solution.

Samples were brought into a field lab and studied in the living state with a wet mount for determination of the general composition of the community. Samples containing more than one group were split into sub-samples because different methods were used for processing and identification. Such samples were preserved in Lugol's Iodine solution. Various methods were used to analyze the various phytoplankton components.

Identification was made from well-known manuals such as Germain (1981), Patrick and Reimer (1966, 1974), Huberpestolozzi (1930-75), Hustedt (1930), Prescott (1951), Smith (1950), Geitler (1932), Desikachary ((1959), West and Fritsch (1927), Komarek and Fott (1983), Wolle (1894), Boyer (1927), Printz (1962), Ettl (1976), Iyengar and Desikachary (1980), Flint (1949), and Philipose (1967). A complete review of the microalgae taken from the Choctawhatchee River system is given by Prasad and Livingston (1987; An Atlas of diatoms and other algal forms from selected drainage areas in central and north Florida; unpublished report for the Florida Department of Environmental Regulation).

A conversion was made of all phytoplankton data (numbers per species /division). This was carried out using seasonal collections of phytoplankton from various portions of the study area. Regressions were determined from the experimental work (comparisons of the numbers vs. the ash-free dry weight

biomass). The following is the protocol used and the results of the regression analysis:

PHYTOPLANKTON-WEIGHT REGRESSION PROTOCOL

1) Find Phytoplankton samples (Rm 126). All should be labelled for station, date, net number, and net size (25 μ m).

2) Weigh Glass-Fiber filters individually in standard weighing tins. Weigh all filters and pans at least separated three times. Make sure to keep accurate records and individually mark each tin. Include at least five filters and pans to be used as controls (untreated).

3) Pour sample through filter and wash any adhering material from sides of sample vial into filter with distilled water. Pour distilled water alone through five control filters. Make sure to put filters into pans they were originally weighed in.

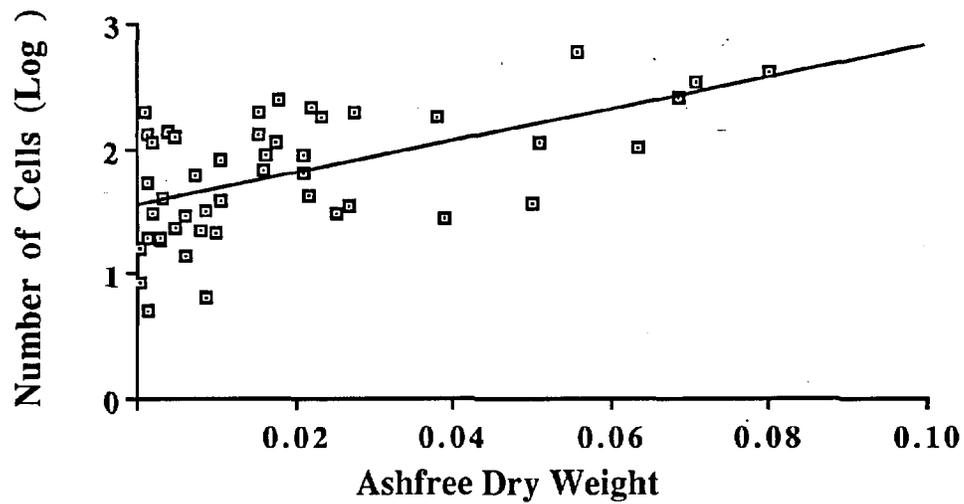
4) Examine filters and remove any detritus or zooplankters present

5) Place filters and pans in drying oven at 95°C-100°C for twentyfour (24) hours. Remove from oven and let cool in Dessicator.

6) Weigh pan/filters on Mettler Balance to lowest possible weight. Be sure all pans are cool before weighing since warm pans will adsorb water from the air and cause the weights to fluctuate. Repeat weights on all pans at least three separate times.

7) Place filter/pans in ashing furnace at 500°C. Burn to ash. Remove filter/pans from oven and let cool in dessicator. Weigh as before.

8) Make sure to keep exact records and follow protocol.



III. PHOTOGRAPHIC ATLAS

The photographic representations of various estuarine algal species are presented in Appendix III.

IV. ANALYSIS OF SPECIES DISTRIBUTION

A. River algae

Although this atlas will concentrate on the estuarine phytoplankton as water quality indicators, a brief description of the river algae is appropriate. A more detailed treatment of the subject is given by Livingston et al. (1988).

A review of the taxonomic organization of the river microalgal components is given in Table 1. In terms of numbers of species and numbers of occurrences in the data base, the Division Bacillariophyta (diatoms) is the most dominant group. In part, this is due to the methods of collection and preservation; such methods favor the collection and analysis of organisms with hardened cell walls such as diatoms. Among the green algae (Division Chlorophyta), the genus *Scenedesmus* is predominant followed by *Closterium* spp. Among the blue-green algae (Cyanophyta), the genus *Merismopedia* is dominant. The genus *Dinobryon* was dominant among the golden-brown algae (Chrysophyta). The euglenoids (Euglenophyta) and cryptomonads (Cryptophyta) were not well represented in these collections. A review of the algae found in the Choctawhatchee region has been given by Prasad and Livingston (1987).

The numbers of microalgae in the main channel stations (70, 68, and 61) were low relative to most of the river tributaries and bay stations. Such low numerical abundance could be a product of the higher flow rates at these stations. Phytoplankton numerical abundance was particularly high in Pine Log Creek, Holmes Creek, Wright's Creek, and the estuarine stations (3, 7, 15).

Table 1: Systematic review of phytoplankton taken with 25 µm nets in the Choctawhatchee River system.

Systematic review

Choctawhatchee River, day, surface, 25µ samples

	No. of occurrences
Division...Cyanophyta	
Class.....Cyanophyceae (blue-green algae or cyanobacteria)	
Order.....Chroococcales	
Family.....Chroococcaceae	
<i>Chroococcus</i> sp.	2
<i>Merismopedia glauco</i>	4
<i>Merismopedia punctata</i>	1
<i>Merismopedia tenuissima</i>	4
Order.....Nostocales	
Family.....Nostocaceae	
<i>Anabaena catenula</i>	1
<i>Anabaena</i> sp.	3
Family.....Oscillatoriaceae	
<i>Arthrospira</i> sp.	1
<i>Oscillatoria amoena</i>	1
<i>Pseudanabaena</i> sp.	1
Division...Euglenophyta	
Class.....Euglenophyceae	
Order.....Euglenales	
<i>Phacus</i> sp. r258	2
<i>Phacus</i> sp.	1
Division...Cryptophyta	
Class.....Cryptophyceae	
<i>Cryptomonas erosa</i>	1
Division...Dinophyta	
Class.....Dinophyceae (dinoflagellates)	
Order.....Gymnodiniales	
Family.....Gymnodiniaceae	
<i>Gymnodinium</i> sp.	1
Order.....Peridinales	
Family.....Peridiniaceae	
<i>Peridinium</i> sp. 2	1
<i>Peridinium</i> sp.	2
Division...Chrysophyta	
Class.....Chrysophyceae (golden-brown algae)	
Order.....Chryomonadales	
Family.....Ochromonadaceae	
<i>Dinobryon sertularia</i>	5
<i>Dinobryon</i> sp. 1	1
Division...Xanthophyta	
Class.....Xanthophyceae (yellow-green algae)	
Order.....Tribonemales	
Family.....Tribonemataceae	
<i>Bumilleria exilis</i>	1
Division...Bacillariophyta (diatoms)	
Class.....Bacillariophyceae	
Order.....Pennales	
Suborder...Araphidineae	
Family.....Diatomaceae	
<i>Asterionella formosa</i>	13

<i>Ctenophora pulchella</i>	1
<i>Fragilaria brevistriata</i>	21
<i>Fragilaria capucina</i>	7
<i>Fragilaria constricta</i>	1
<i>Fragilaria construens</i>	9
<i>Fragilaria leptostauron v. dubia</i>	1
<i>Fragilaria elliptica</i>	2
<i>Fragilaria leptostauron</i>	3
<i>Fragilaria pinnata</i>	27
<i>Fragilaria construens v. venata</i>	7
<i>Fragilaria construens v. venter</i>	13
<i>Meridion circulare</i>	4
<i>Opephora americana</i>	2
<i>Opephora gemmata</i>	1
<i>Opephora martyi</i>	16
<i>Opephora pacifica</i>	1
<i>Opephora pinnata</i>	3
<i>Opephora schwartzii</i>	6
<i>Pleurosira laevis</i>	2
<i>Rhabdonema adriaticum</i>	3
<i>Synedra sp. 265</i>	2
<i>Synedra acus</i>	20
<i>Synedra ulna v. amphirhynchus</i>	5
<i>Synedra delicatissima</i>	9
<i>Synedra filiformis</i>	3
<i>Synedra incisa</i>	1
<i>Synedra ulna v. oxyrhynchus</i>	19
<i>Synedra rumpens</i>	2
<i>Synedra sp. 1</i>	1
<i>Synedra sp.</i>	1
<i>Synedra tabulata</i>	3
<i>Synedra ulna</i>	75
<i>Tabellaria binalis</i>	1
<i>Tabellaria fenestrata</i>	26
<i>Tabellaria flocculosa</i>	15
<i>Tabellaria quadrisepata</i>	19
<i>Tabularia investiens</i>	7
Suborder...Raphidineae	
Family.....Eunotiaceae	
<i>Actinella punctata</i>	3
<i>Eunotia alpina</i>	28
<i>Eunotia monodon v. major f. bidens</i>	3
<i>Eunotia bigibba</i>	1
<i>Eunotia diodon</i>	4
<i>Eunotia exigua</i>	1
<i>Eunotia faba</i>	2
<i>Eunotia flexuosa</i>	6
<i>Eunotia formica</i>	22
<i>Eunotia lunaris</i>	12
<i>Eunotia monodon v. major</i>	1
<i>Eunotia monodon</i>	15
<i>Eunotia naegeli</i>	3
<i>Eunotia pectinalis</i>	89
<i>Eunotia polydentula</i>	1
<i>Eunotia praerupta</i>	7
<i>Eunotia praerupta v. bidens</i>	1
<i>Eunotia pectinalis v. undulata</i>	20
<i>Eunotia purpussilla</i>	1
<i>Eunotia quaternaris</i>	1
<i>Eunotia serra</i>	6

<i>Eunotia</i> sp. 2	1
<i>Eunotia</i> sp. 3	1
<i>Eunotia</i> sudetica	1
<i>Eunotia</i> tautoniensis	5
<i>Eunotia</i> pectinalis v. ventricosa	1
<i>Eunotia</i> zygodon	1
<i>Semiorbis</i> hemicyclus	1
Family.....Achnanthaceae	
<i>Achnanthes</i> affinis	4
<i>Achnanthes</i> lanceolata v. apiculata	81
<i>Achnanthes</i> austriaca	3
<i>Achnanthes</i> bottanica	1
<i>Achnanthes</i> clevei	33
<i>Achnanthes</i> conspicua	3
<i>Achnanthes</i> delicatula	7
<i>Achnanthes</i> lanceolata v. dubia	65
<i>Achnanthes</i> lanceolata v. elliptica	8
<i>Achnanthes</i> exigua	39
<i>Achnanthes</i> flexella	20
<i>Achnanthes</i> peragalli v. fossilis	1
<i>Achnanthes</i> haukiana	2
<i>Achnanthes</i> lanceolata v. haynaldii	1
<i>Achnanthes</i> exigua v. heterovalva	2
<i>Achnanthes</i> hustedtii	9
<i>Achnanthes</i> inflata	1
<i>Achnanthes</i> lanceolata v. lanceolatoides	10
<i>Achnanthes</i> lanceolata	12
<i>Achnanthes</i> linearis	1
<i>Achnanthes</i> lanceolata v. omissa	17
<i>Achnanthes</i> peragalli v. parvula	2
<i>Achnanthes</i> peragallii	19
<i>Achnanthes</i> lanceolata v. rostrata	1
<i>Achnanthes</i> saxonica	1
<i>Achnanthes</i> sp. 1	1
<i>Cocconeis</i> diminuta	15
<i>Cocconeis</i> disculus	5
<i>Cocconeis</i> pediculus	1
<i>Cocconeis</i> placentula	70
Family.....Naviculaceae	
<i>Amphipleura</i> pellucida	1
<i>Amphora</i> disculus	1
<i>Amphora</i> ovalis	11
<i>Amphora</i> pediculus	3
<i>Anomoeneis</i> brachysira	7
<i>Anomoeneis</i> serians	5
<i>Anomoeneis</i> serians v. brachysira	1
<i>Anomoeneis</i> vitrea	1
<i>Brachysira</i> apomina	1
<i>Caloneis</i> molaris	1
<i>Capartogramma</i> crucicula	49
<i>Cymbella</i> affinis	7
<i>Cymbella</i> amphicephala	2
<i>Cymbella</i> aspera	11
<i>Cymbella</i> cesatii	1
<i>Cymbella</i> cistula	7
<i>Cymbella</i> cuspidata	4
<i>Cymbella</i> delicatula	1
<i>Cymbella</i> gracilis	20
<i>Cymbella</i> helvetica	3
<i>Cymbella</i> hustedii	1

<i>Cymbella lapponica</i>	1
<i>Cymbella leptoceros</i>	1
<i>Cymbella lunata</i> v. <i>lunata</i>	1
<i>Cymbella minuta</i>	4
<i>Cymbella naviculiformis</i>	5
<i>Cymbella</i> sp.1	1
<i>Cymbella subcuspidata</i>	35
<i>Cymbella tumida</i>	13
<i>Cymbella turgidula</i>	1
<i>Cymbella ventricosa</i>	1
<i>Diploneis elliptica</i>	5
<i>Diploneis</i> sp.	1
<i>Diploneis oblongella</i>	1
<i>Diploneis ovalis</i>	3
<i>Diploneis parma</i>	3
<i>Frustulia rhomboides</i> v. <i>crassinerva</i>	1
<i>Frustulia rhomboides</i>	83
<i>Frustulia rhomboides</i> v. <i>saxonica</i>	1
<i>Frustulia vulgaris</i>	5
<i>Gomphonema acuminatum</i>	3
<i>Gomphonema angustatum</i>	35
<i>Gomphonema augur</i>	1
<i>Gomphonema gracile</i>	12
<i>Gomphonema grovei</i>	4
<i>Gomphonema intricatum</i>	1
<i>Gomphonema grovei</i> v. <i>lingulatum</i>	7
<i>Gomphonema minima</i>	1
<i>Gomphonema parvulum</i>	3
<i>Gomphonema truncatum</i>	2
<i>Gyrosigma acuminatum</i>	27
<i>Gyrosigma attenuatum</i>	5
<i>Gyrosigma kuetzingii</i>	1
<i>Gyrosigma scalproides</i>	2
<i>Gyrosigma spenceri</i>	16
<i>Navicula</i> sp. 248	1
<i>Navicula</i> sp. 251	1
<i>Navicula</i> cf. <i>ammophila</i>	1
<i>Navicula anglica</i>	4
<i>Navicula bacillaris</i>	1
<i>Navicula bacillum</i>	1
<i>Navicula begerii</i>	1
<i>Navicula capitata</i>	26
<i>Navicula clementis</i>	8
<i>Navicula clementoides</i>	1
<i>Navicula contenta</i>	1
<i>Navicula cohnii</i>	1
<i>Navicula constans</i>	12
<i>Navicula capitata</i> v. <i>capitata</i>	2
<i>Navicula cryptocephala</i>	66
<i>Navicula cuspidata</i>	7
<i>Navicula cryptonella</i>	1
<i>Navicula decussis</i>	21
<i>Navicula dicephala</i>	21
<i>Navicula disculus</i>	2
<i>Navicula elginense</i>	1
<i>Navicula gastrum</i>	11
<i>Navicula gracilis</i>	1
<i>Navicula gregaria</i>	1
<i>Navicula halophila</i>	7
<i>Navicula hasta</i>	16

<i>Navicula hungarica</i>	5
<i>Navicula cryptocephala v. intermedia</i>	1
<i>Navicula jaernefeltii</i>	2
<i>Navicula lacustris</i>	1
<i>Navicula laevissima</i>	17
<i>Navicula leptostriata</i>	3
<i>Navicula lucinensis</i>	1
<i>Navicula capitata v. lueneburgensis</i>	2
<i>Navicula menisculus</i>	2
<i>Navicula minima</i>	2
<i>Navicula placenta</i>	4
<i>Navicula placentula</i>	6
<i>Navicula protracta</i>	4
<i>Navicula psuedoscutiformis</i>	6
<i>Navicula pupula</i>	41
<i>Navicula radiosa</i>	20
<i>Navicula rhyncocephala</i>	10
<i>Navicula salsa</i>	3
<i>Navicula subplacentula</i>	1
<i>Navicula subrhyncocephala</i>	1
<i>Navicula schroeteri</i>	11
<i>Navicula scutelloides</i>	2
<i>Navicula sp. 5</i>	1
<i>Navicula sp. 6</i>	1
<i>Navicula sp. 7</i>	1
<i>Navicula sp. 8</i>	1
<i>Navicula sp. 9</i>	1
<i>Navicula sp. 10</i>	1
<i>Navicula sp.</i>	2
<i>Navicula scutiformis</i>	2
<i>Navicula stroemii</i>	1
<i>Navicula subhamulata</i>	1
<i>Navicula tanera</i>	2
<i>Navicula trivialis</i>	4
<i>Navicula tuscula</i>	2
<i>Navicula viridula</i>	66
<i>Navicula radiosa v. tenella</i>	1
<i>Navicula yarrensensis</i>	1
<i>Navicula yecens</i>	1
<i>Neidium affine</i>	19
<i>Neidium affinis v. amphyrineus</i>	1
<i>Neidium ampliatum</i>	2
<i>Neidium binode</i>	1
<i>Neidium densestriatum</i>	1
<i>Neidium dilatatum</i>	2
<i>Neidium dubium</i>	6
<i>Neidium iridis</i>	12
<i>Neidium productum</i>	5
<i>Pinnularia acrosphaeria</i>	1
<i>Pinnularia acuminata</i>	1
<i>Pinnularia borealis</i>	3
<i>Pinnularia braunii</i>	2
<i>Pinnularia divergens</i>	1
<i>Pinnularia gibba</i>	28
<i>Pinnularia legumen</i>	4
<i>Pinnularia major</i>	1
<i>Pinnularia microstauron</i>	5
<i>Pinnularia obscura</i>	2
<i>Pinnularia sp. 2</i>	1
<i>Pinnularia sp. 3</i>	1

<i>Pinnularia subcapitata</i>	4
<i>Pinnularia sudetica</i>	5
<i>Pinnularia viridis</i>	12
<i>Stauroneis agrestis</i>	1
<i>Stauroneis anceps</i>	17
<i>Stauroneis anceps f. gracilis</i>	3
<i>Stauroneis nobilis</i>	1
<i>Stauroneis phoenicenteron</i>	17
<i>Stauroneis prominula</i>	1
<i>Stauroneis schimanskii</i>	1
<i>Stauroneis anceps v. siberica</i>	1
<i>Stauroneis smithii</i>	16
<i>Stauroneis sp. 2</i>	1
<i>Stauroneis sp. 3</i>	1
<i>Tropidoneis lepidoptera</i>	3
Family.....Epithemiaceae	
<i>Epithemia zebra v. porcellum</i>	1
<i>Epithemia zebra</i>	12
<i>Rhopalodia gibba</i>	2
<i>Rhopalodia gibberula</i>	1
Family.....Nitzschiaceae	
<i>Bacillaria paxillifer</i>	28
<i>Denticula sp.</i>	1
<i>Denticula thermalis</i>	1
<i>Hantzschia amphioxys</i>	1
<i>Hantzschia virgata v. capitellata</i>	3
<i>Hantzschia virgata</i>	8
<i>Nitzschia acicularis</i>	18
<i>Nitzschia acuta</i>	4
<i>Nitzschia amphibia</i>	3
<i>Nitzschia acicularis v. closterioides</i>	2
<i>Nitzschia communis</i>	2
<i>Nitzschia closterium</i>	1
<i>Nitzschia dissipata</i>	6
<i>Nitzschia dubia</i>	2
<i>Nitzschia frugalis</i>	3
<i>Nitzschia gracilis</i>	8
<i>Nitzschia hantzschiana</i>	3
<i>Nitzschia tryblionella v. laevidensis</i>	4
<i>Nitzschia linearis</i>	1
<i>Nitzschia tryblionella v. levidensis</i>	2
<i>Nitzschia palaecea</i>	3
<i>Nitzschia palea</i>	21
<i>Nitzschia parvula</i>	1
<i>Nitzschia romana</i>	12
<i>Nitzschia rostellata</i>	1
<i>Nitzschia sigmoidea</i>	6
<i>Nitzschia sp. 3</i>	2
<i>Nitzschia sp. 4</i>	1
<i>Nitzschia sp. 5</i>	2
<i>Nitzschia sp. 6</i>	2
<i>Nitzschia sp. 7</i>	1
<i>Nitzschia tryblionella v. subsalina</i>	1
<i>Nitzschia triblionella</i>	24
<i>Nitzschia tryblionella v. victoriae</i>	4
<i>Nitzschia vitrea-like</i>	1
Family.....Surirellaceae	
<i>Campylodiscus sp.</i>	1
<i>Cymatopleura solea</i>	2
<i>Stenopterobia intermedia</i>	2

<i>Surirella angustata</i>	5
<i>Surirella biseriata</i>	6
<i>Surirella elegans</i>	3
<i>Surirella gracilis</i>	4
<i>Surirella linearis</i>	16
<i>Surirella ovata</i>	5
<i>Surirella robusta</i>	15
<i>Surirella tenera</i>	59
Order.....Centrales	
Suborder...Coscinodiscineae	
Family.....Thalassiosiraceae	
<i>Aulacosira granulata</i>	16
<i>Cyclotella meneghiniana</i>	21
<i>Cyclotella stelligera</i>	1
<i>Cyclotella striata</i>	6
<i>Thalassiosira decipiens</i>	1
<i>Thalassiosira eccentrica</i>	1
<i>Thalassiosira sp. 1</i>	1
<i>Thalassiosira sp. 2</i>	1
Family.....Melosiraceae	
<i>Melosira italica</i>	1
<i>Melosira undulata</i>	35
<i>Melosira varians</i>	31
<i>Paralia sulcata</i>	1
Suborder...Biddulphiineae	
Family.....Biddulphiaceae	
<i>Hydrosera triquetia</i>	2
<i>Terpsinoe americana</i>	2
<i>Terpsinoe musica</i>	3
Division...Chlorophyta	
Class.....Chlorophyceae (green algae)	
Order.....Chaetophorales	
Family.....Chaetophoraceae	
<i>Chaetophora-like form</i>	1
Order.....Chlorococcales	
Family.....Hydrodictyaceae	
<i>Pediastrum duplex</i>	1
<i>Pediastrum simplex</i>	2
Family.....Scenedesmaceae	
<i>Scenedesmus sp. 252</i>	1
<i>Scenedesmus armatus</i>	6
<i>Scenedesmus dimorphus</i>	12
<i>Scenedesmus obliquus</i>	3
<i>Tetraedron trigonium v. gracile</i>	1
Order.....Volvocales	
Family.....Volvocaceae	
<i>Pandorina morum</i>	3
Order.....Zygnemales	
Family.....Desmediaceae	
<i>Arthrodesmus octocorne</i>	2
<i>Arthrodesmus subulatus</i>	1
<i>Arthrodesmus triangularis v.</i>	1
<i>Bambusina brebissonii</i>	2
<i>Closterium bailyanum</i>	4
<i>Closterium libellula</i>	3
<i>Closterium sp. 1</i>	3
<i>Closterium sp. 2</i>	1
<i>Closterium sp. 3</i>	2
<i>Closterium sp. 4</i>	3

<i>Closterium</i> sp. 5 (giant one r254/29)	1
<i>Cosmarium contractum</i>	1
<i>Cosmarium punctulatum</i>	1
<i>Cosmarium</i> sp. 1	1
<i>Cosmarium</i> sp. 2 (254)	1
<i>Cylindrocystis americana</i>	1
<i>Desmidium aptogonum</i>	1
<i>Docidium undulatum</i>	1
<i>Euastrum affine</i>	1
<i>Euastrum gemmatum</i>	1
<i>Gonatozygon pilosum</i>	1
<i>Gymnozyga moniliformis</i>	1
<i>Penium</i> sp.	1
<i>Staurastrum cornatum</i>	2
<i>Staurastrum limneticum</i> v. <i>cornutum</i>	1
<i>Staurastrum curvatum</i>	2
<i>Staurastrum paradoxum</i>	3
<i>Staurastrum cornatum</i> s. <i>cornutum</i>	1
<i>Staurastrum</i> sp. 1	1
<i>Tetmemorus brebissonii</i>	2
<i>Tetmemorus granulatus</i>	1
Family.....Zygnemaceae	
<i>Mougeotia elongatula</i>	1
<i>Mougeotia</i> sp. 1 (narrow trichomes)	2
<i>Mougeotia</i> sp. 2 (wide trichomes)	2
<i>Mougeotia</i> sp. 3	1

Species richness in the main stream was comparable to that in the various tributaries. The species richness in most of the river stations was considerably higher than that in the estuary. The data indicate that main stream phytoplankton abundance and species richness showed relative stereotypic distributions in terms of the overall habitat characteristics in the river-estuarine system. The uniformly high numbers of phytoplankton in the productive estuary throughout the year was in contrast to the more seasonal phytoplankton abundance in the river. Areas of high flow were characterized by low numbers of individuals; species richness in the main stem, however, was comparable to the tributaries. The salinity-stressed estuarine conditions could be associated with the low species richness in the eastern portion of Choctawhatchee Bay.

In terms of species richness at most of the river stations, there was a general pattern of high numbers of species during January, 1987 followed by a decline during the succeeding months with or without a second peak during the fall. Overall species richness for a given month was highest in Holmes Creek. Species richness patterns in the estuary were quite variable from station to station. At station 15, there were peaks during late spring and winter months. At stations 3 and 7, such peaks tended to occur during spring and summer months. In terms of numerical abundance, there was considerable station to station variability. In general, there were winter-spring and fall peaks of abundance, but such peaks followed station-specific patterns over the period of observation. The high numbers of phytoplankton in Pine Log Creek (due to the February bloom) and Holmes Creek (again, a February bloom contributed heavily to the observed winter peak) are evident in contrast to the very low numbers of phytoplankton taken at the upper Choctawhatchee River station and in Bruce Creek and Sandy Creek. The biomass trends followed the numerical

trends closely. When viewed as mean numbers of species (cumulative species richness), another pattern is evident. The lowest phytoplankton mean species richness occurred in Seven Mile Creek (62) followed closely by Bruce Creek (64) and the upper Choctawhatchee River (70). The highest mean species richness of phytoplankton in the Choctawhatchee system occurred in Pine Log Creek (60) and Sandy Creek. The other main stem stations and larger Creeks (Holmes, Wright's) had intermediate levels of cumulative species richness.

Over the period of study, the main channel stations (70,68,61) had relatively high species-specific dominance with 6-8 species accounting for the overall numerical abundance of phytoplankton over the year of study. A group of 7-13 sub-dominant species were present followed in order of abundance by a considerable number of species with low numbers taken over the study period. Species such as *Achnanthes lanceolata* v. *dubia* and v. *apiculata*, *Navicula* spp, *Surirella tenera*, and *Synedra ulna* were usually the top dominants at the main (channel) stations. Overall (cumulative) totals of the numbers of species for the year tended to increase downstream (station 70, 81; station 68, 100; station 61, 101).

In Wright's Creek (station 69), the dominance patterns in terms of numbers of individual species populations tended to follow that described at the main river stations. There were some differences in the sub-dominant species. The cumulative total number of species in Wright's Creek was 114. In Holmes Creek (station 65), dominance hierarchies were quite different than those described above. Top dominants included species such as *Achnanthes clevei*, *Cocconeis placentula* and *C. dimunuta*, *Eunotia pectinalis*, *Fragilaria constuensis*, and the *Achnanthes* spp. There was a cumulative total of 128

species in Holmes Creek. In Sandy Creek (station 67), the top dominants were fewer in number although the species strongly resemble those described for the main river stations and Wright's Creek. The numbers were quite low in this creek and the cumulative species total was 112, similar to that in Wright's Creek. Bruce Creek (station 64) showed a somewhat different pattern with a single top dominant, *Eunotia pectinalis*, followed by a series of 11 sub-dominants that resembled the species lists described above for the various main channel stations. The cumulative total of 123 species was on the high side resembling that of Holmes Creek.

In Seven Run Creek (station 62), the pattern of species abundance resembled that described for Bruce Creek with *Eunotia pectinalis* as the top dominant followed by a series of sub-dominants composed of species similar to those described above for the various other stations. Once again, numbers of species were relatively low in this creek, with a cumulative total of only 94 species. In Pine Log Creek, the dominance pattern followed that described for Bruce Creek and Seven Run Creek. A cumulative total of 145 species at this station was the highest such number found in the survey. The phytoplankton data in Pine Log Creek (station 60) indicate a similar pattern to that described for some of the tributaries. *Eunotia pectinalis* was the top dominant followed by a group of sub-dominants that included most of the familiar species listed as sub-dominants at the various other river stations. The numbers at this station were periodically high so that substantial totals were noted for *Achnanthes* spp, *Frustulia rhomboides*, *Navicula* spp, and *Eunotia alpina*. The cumulative species richness at the various river stations follows a pattern that is similar to though not identical with the mean species numbers (means of the numbers of species per sample). The cumulative species number was highest at Pine Log

Chaetoceros spp. The high numbers of phytoplankton per month were accompanied by a relatively low cumulative number of species (92). At station 7, the top dominant was the same as that described at station 3; the sub-dominants were similar in terms of species and distribution although the exact order within the dominance hierarchy was somewhat different. Overall numbers were substantially higher at station 7; the cumulative species richness was 97. At station 15, the numbers were higher still with the top dominant being *Cyclotella striata*. The sub-dominants were somewhat different in terms of species than those described above. The cumulative species richness of 87 was relatively low. Thus, compared with the river stations, the estuarine areas had higher numbers of phytoplankton though lower cumulative species richness than the river stations. Dominance was similar among the various bay stations although the species noted were quite different from those observed at the river stations.

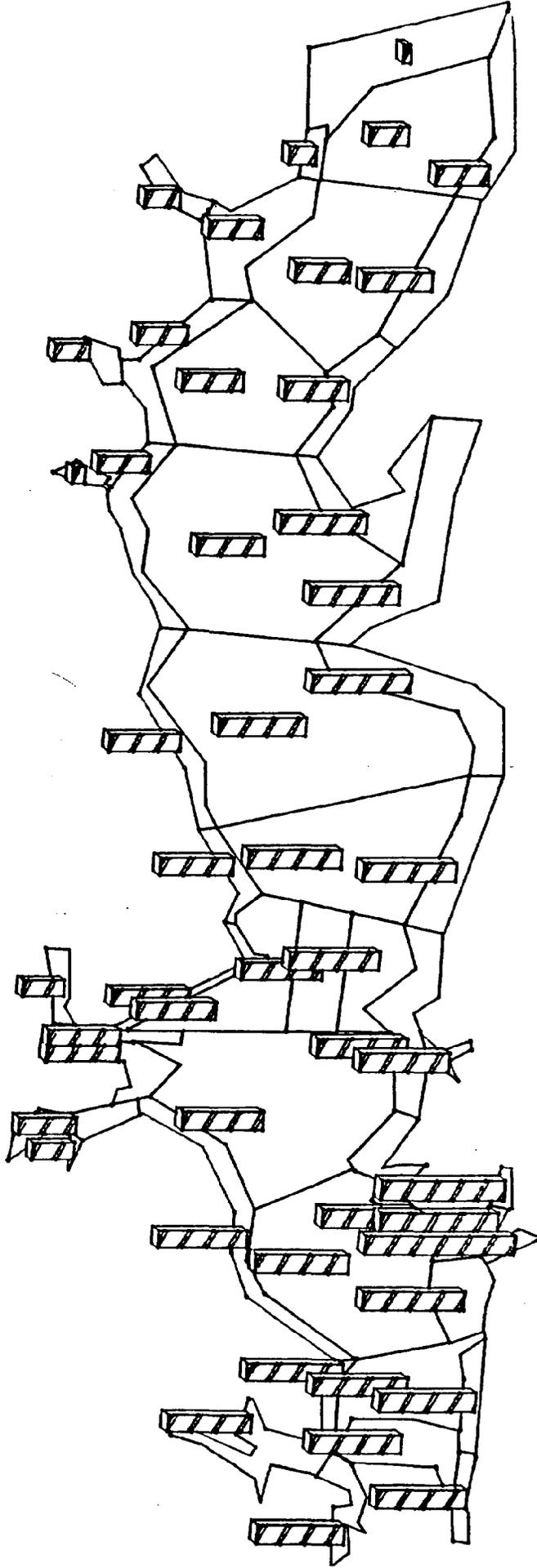
B. Estuarine phytoplankton distribution

1. Water quality

A summary review of water quality is given in Figure 2. A detailed analysis of these data is given in Livingston (1986a). Salinity in the Choctawhatchee Bay system is lowest in eastern sections due to river input and in the northern bayous. Salinity was high in Old Pass Lagoon. Peak salinities occurred in the bay during the months of July through October. Vertical salinity stratification occurs in various portions of the estuary during certain months. Such stratification is evident in the vertical dissolved oxygen distribution that occurs in the bay during warm months. Deep stations throughout the bay had

CHOCTAWHATCHEE BAY

Salinity in ppt
Average Surface Samples For All Dates



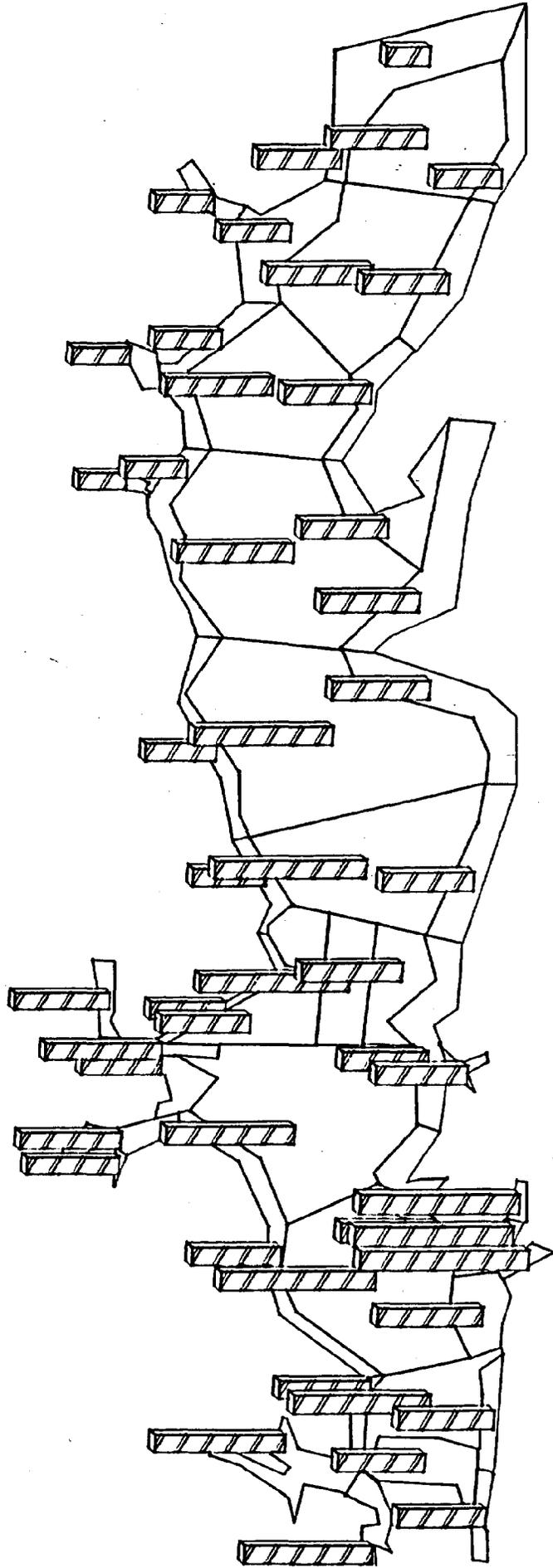
SCALED

0 to 32 by .1

CHOCTAWHATCHEE BAY

Salinity in ppt

Average Bottom Samples For All Dates

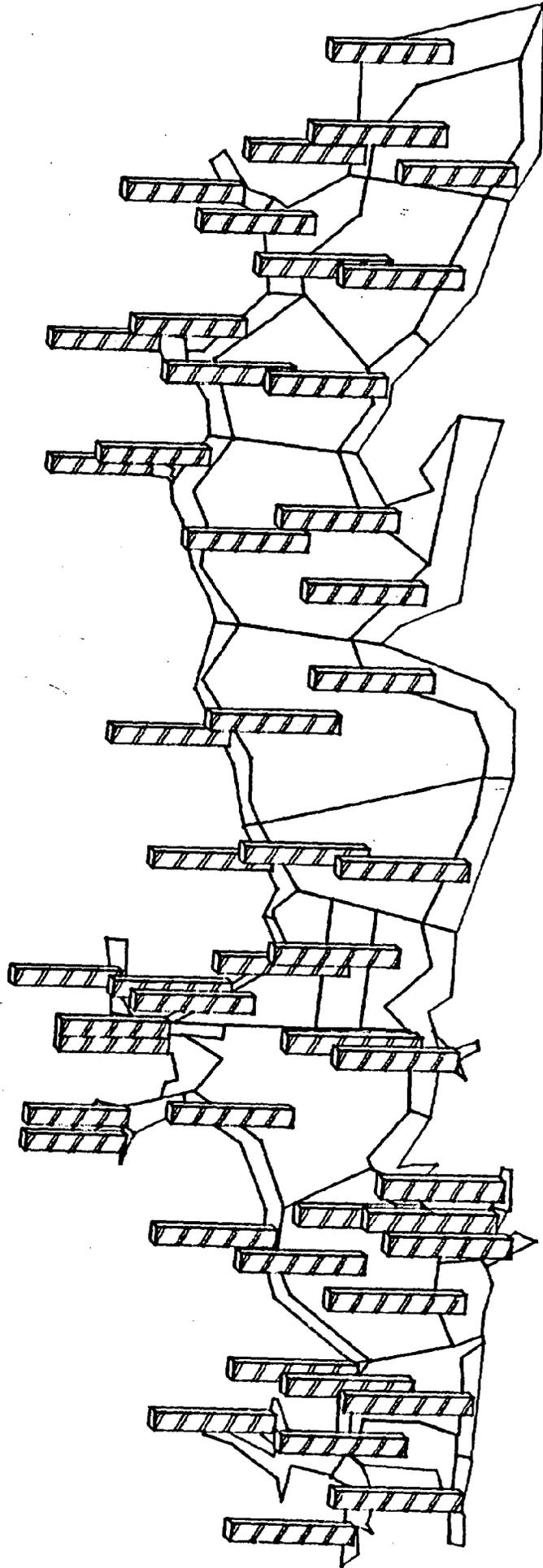


SCALED

0 to 30 ppt

CHOCTAWHATCHEE BAY

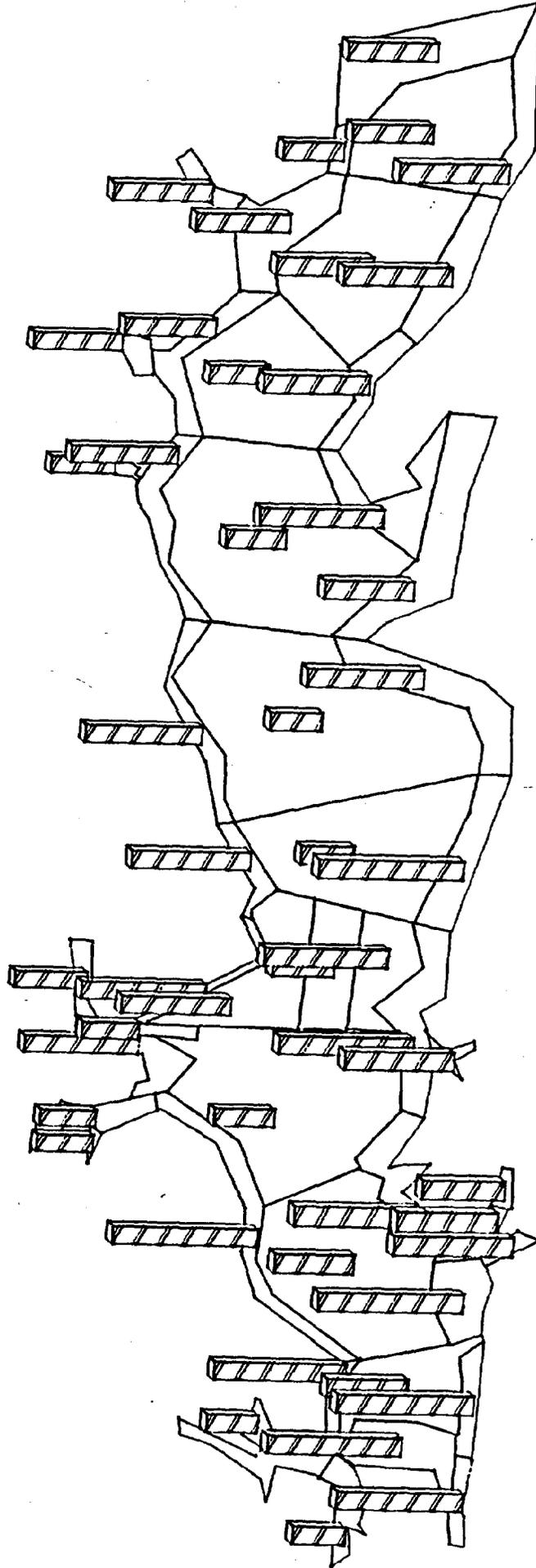
Dissolved Oxygen in ppm
Average Surface Samples For All Dates



SCALED
1 to 10 by .5

CHOCTAWHATCHEE BAY

Dissolved Oxygen in ppm
Average Bottom Samples For All Dates

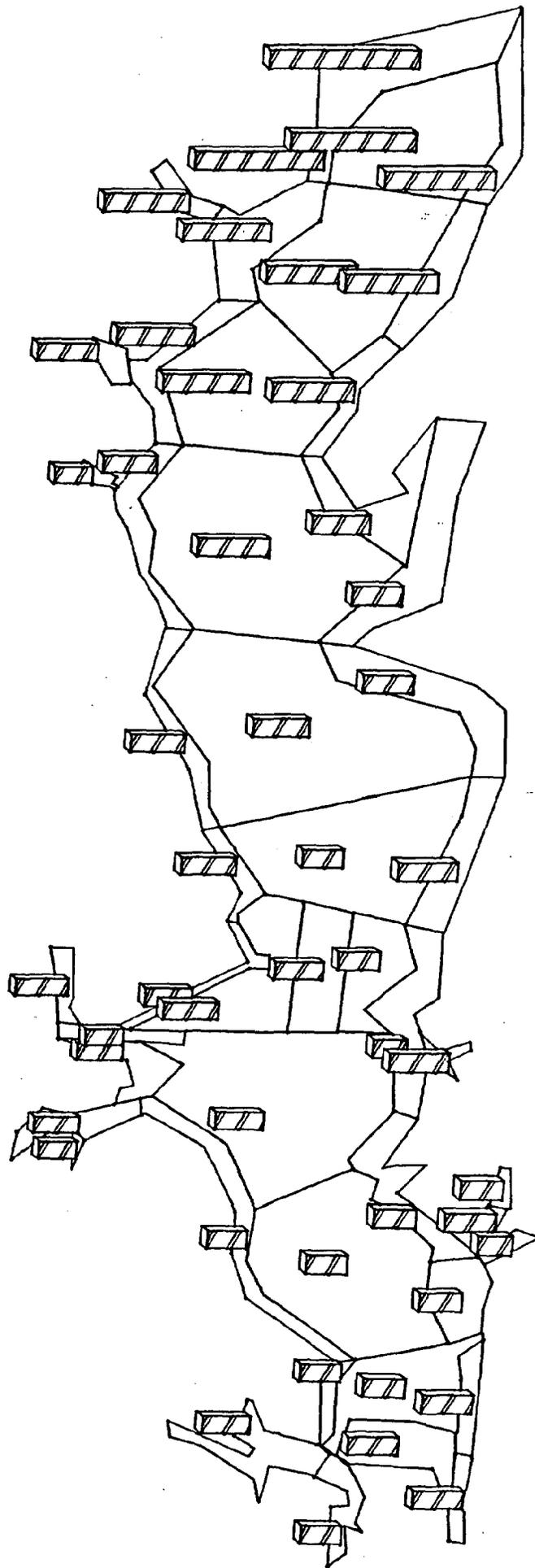


SCALED
1 to 10 by .5

CHOCTAWHATCHEE BAY

Turbidity in NTU

Average Surface Samples For All Dates

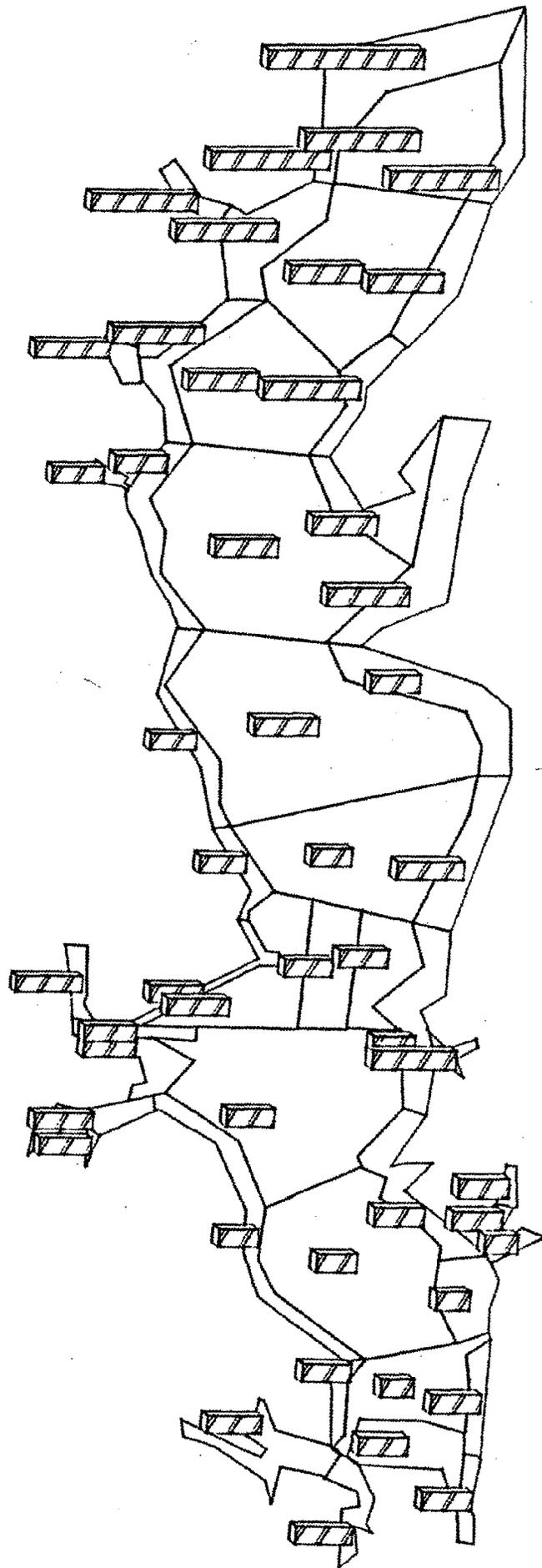


SCALED

0 to 10 by .5

CHUCIAWHAICHEE BAY

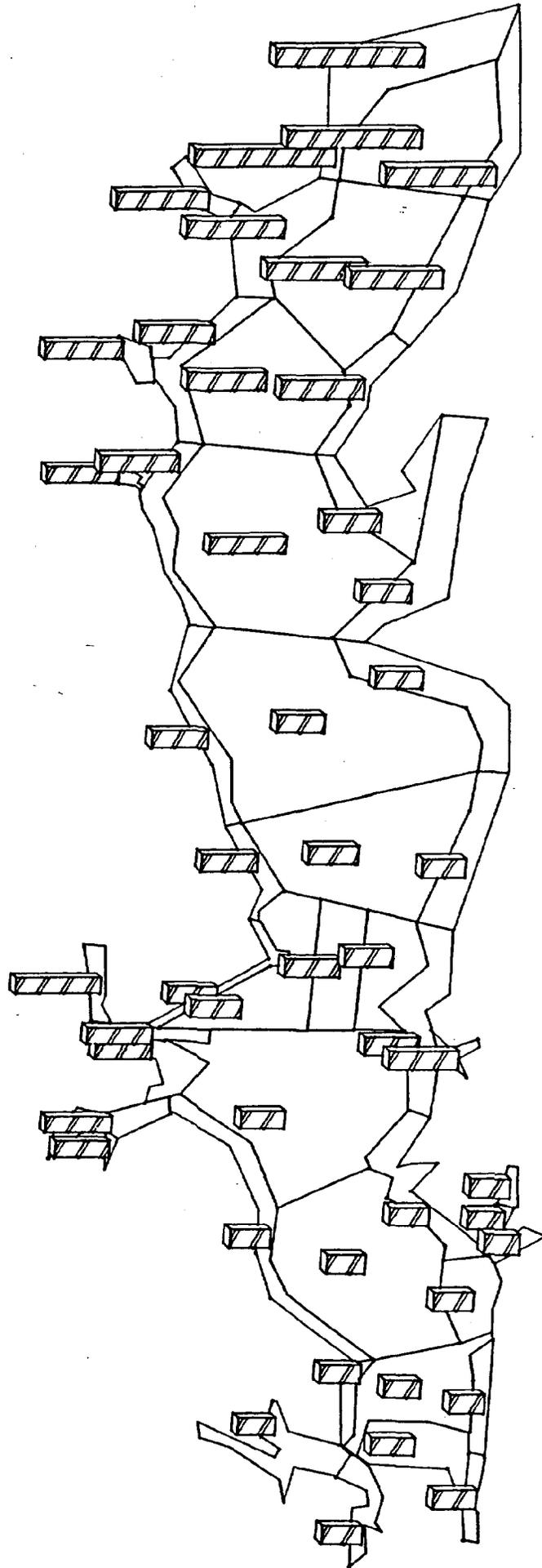
Turbidity in NTU
Average Bottom Samples For All Dates



SCALED
0 to 10 by .5

CHOCTAWHATCHEE BAY

Color in Pt-Co Units
Average Surface Samples For All Dates



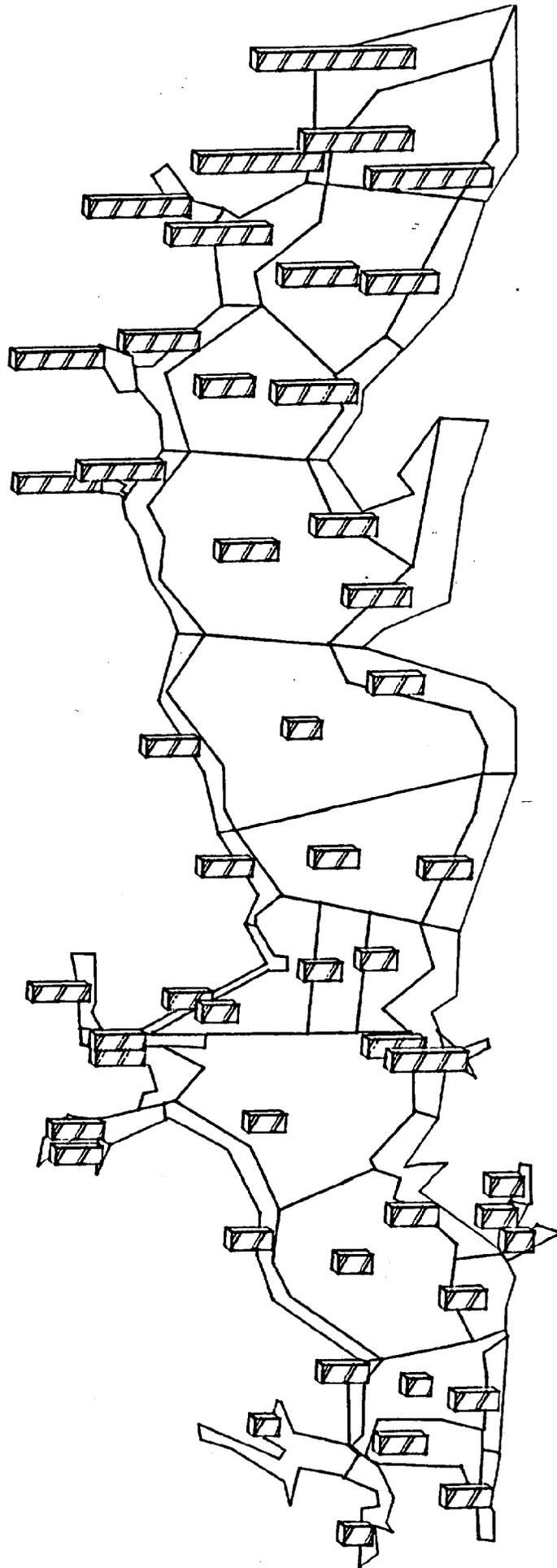
SCALED

0 to 100 %

CHOCTAWHATCHEE BAY

Color in Pt-Co Units

Average Bottom Samples For All Dates

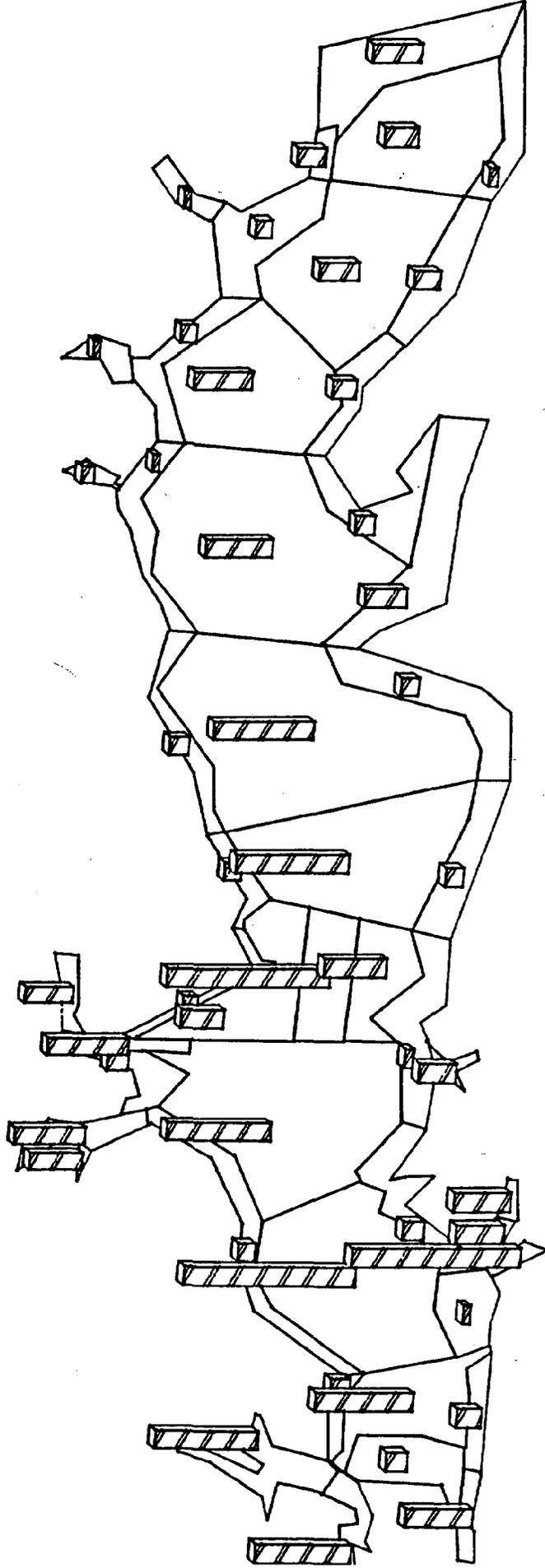


SCALED

0 to 100 by 1

CHOCTAWHATCHEE BAY

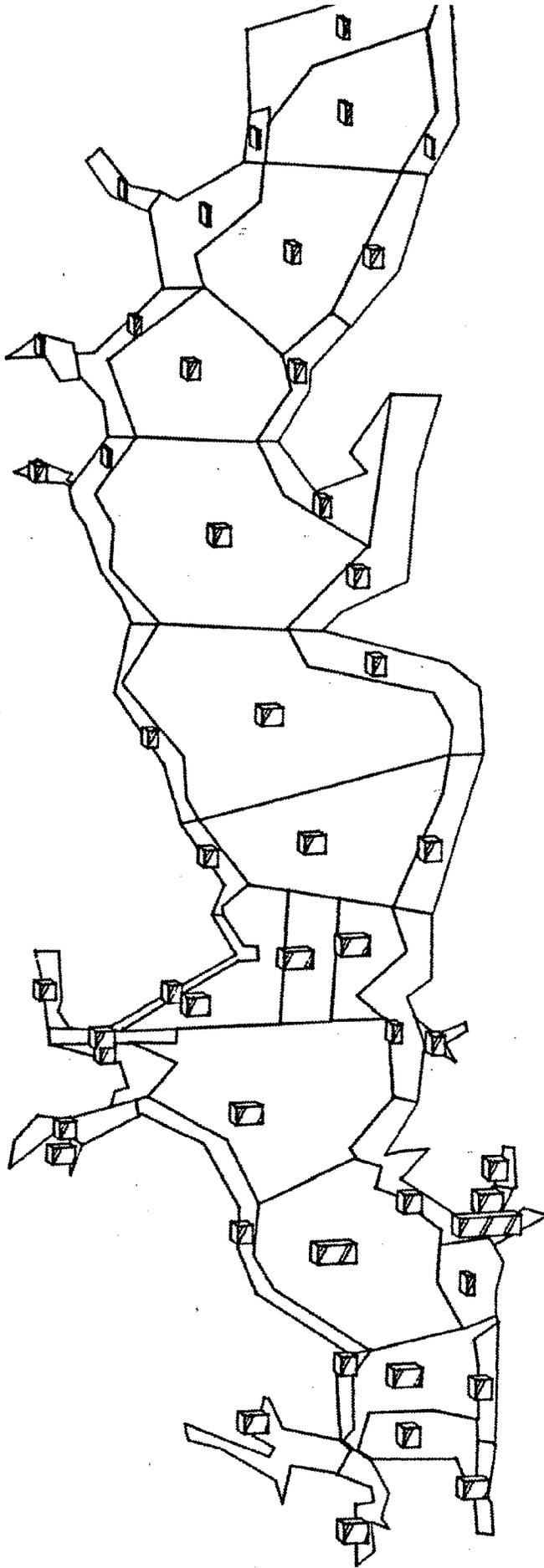
Depth in Meters
Average of All Sample Dates



SCALED
1 to 11 by -1

CHOCTAWHATCHEE BAY

Secchi Readings in Meters
Average of All Sample Dates

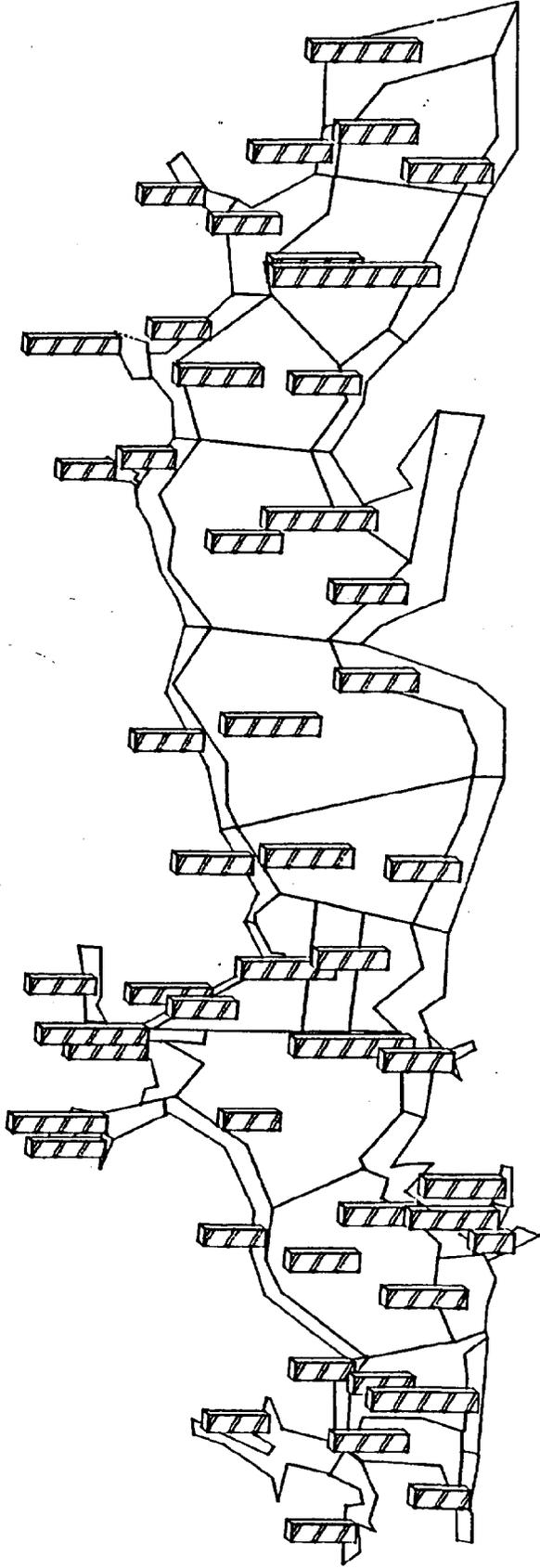


SCALED

1 to 11 by -1

CHOCTAWHATCHEE BAY

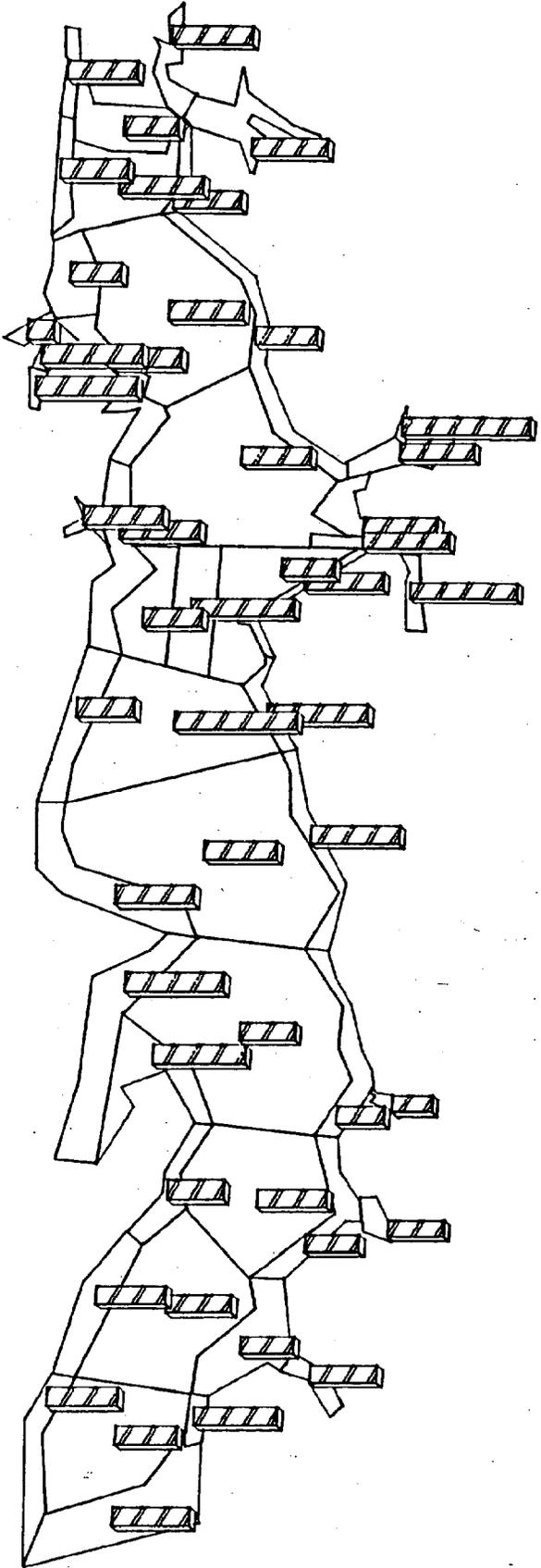
Total Nitrogen in Milligrams Per Liter
Average of Surface Samples For All Dates



0 to 1.2 by .01

CHOCTAWHATCHEE BAY

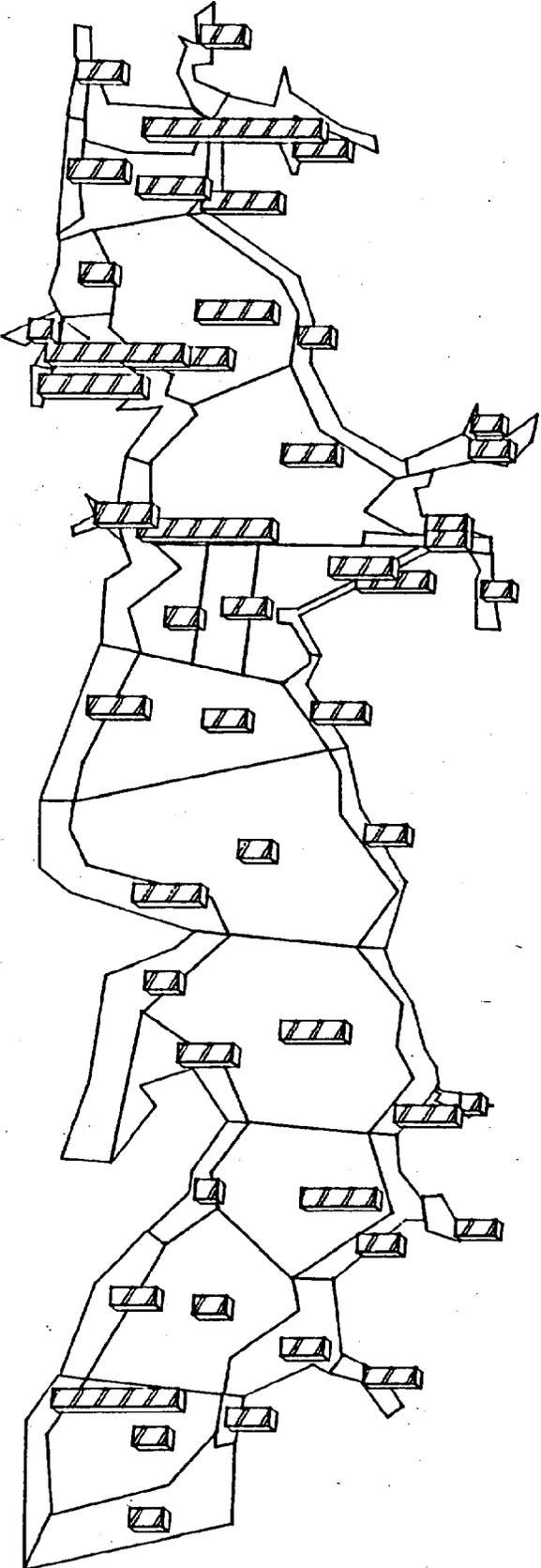
Total Kjeldahl Nitrogen in Milligrams Per Liter
Average of Bottom Samples For All Dates



0 to 1.11 by .01

CHOCTAWHATCHEE BAY

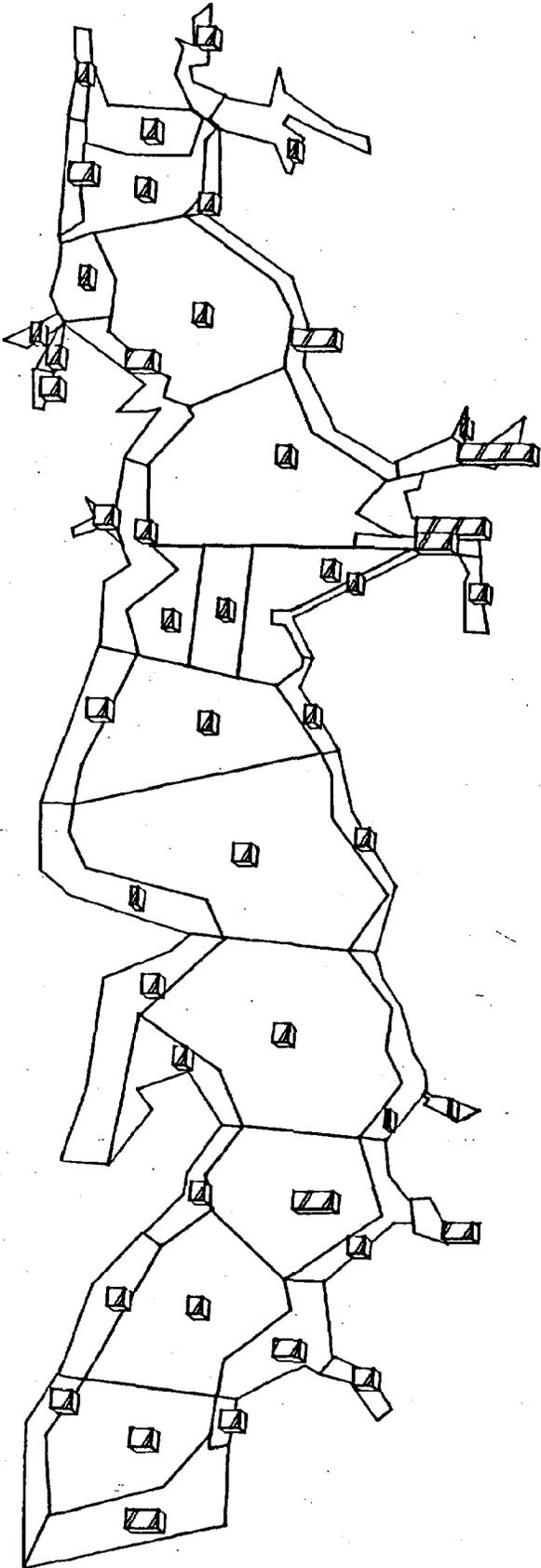
Ortho-phosphate in Milligrams Per Liter
Average of Bottom Samples For All Dates



0 to .015 by .001
Highest value should be .025
@ Sta 42

CHOCTAWHATCHEE BAY

Total Phosphate in Milligrams Per Liter
Average of Surface Samples For All Dates

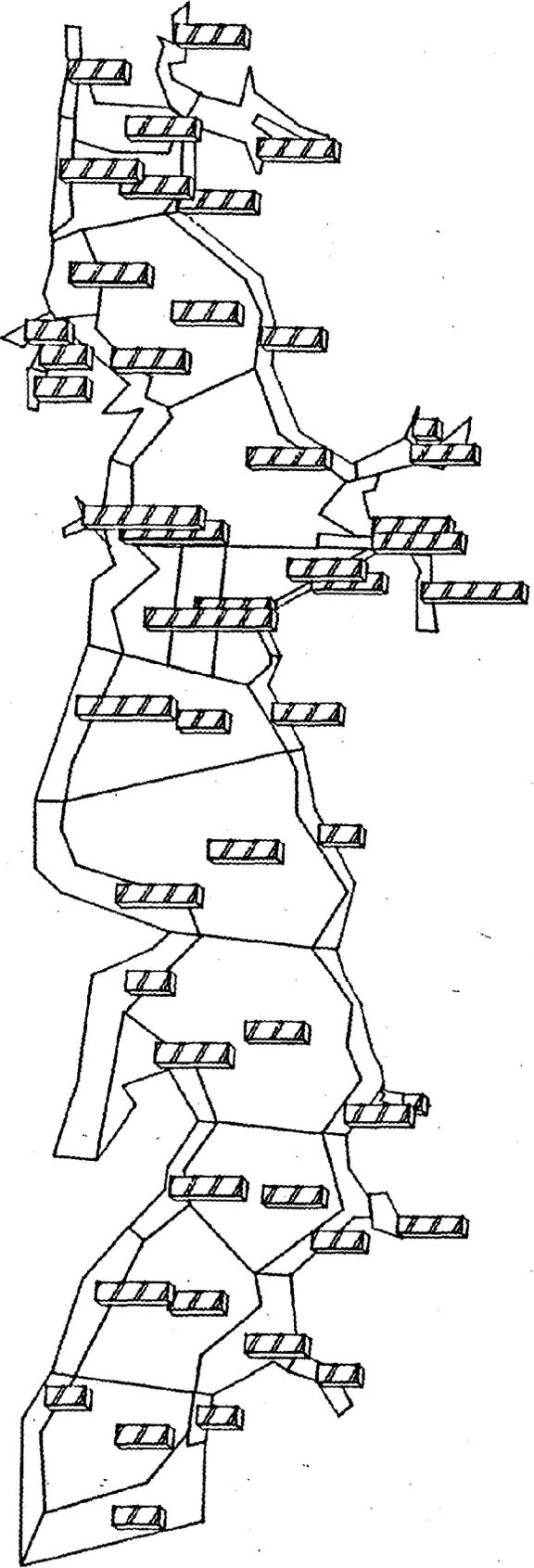


0 to .095 by .001

CHOCTAWHATCHEE BAY

Ammonia in Milligrams Per Liter

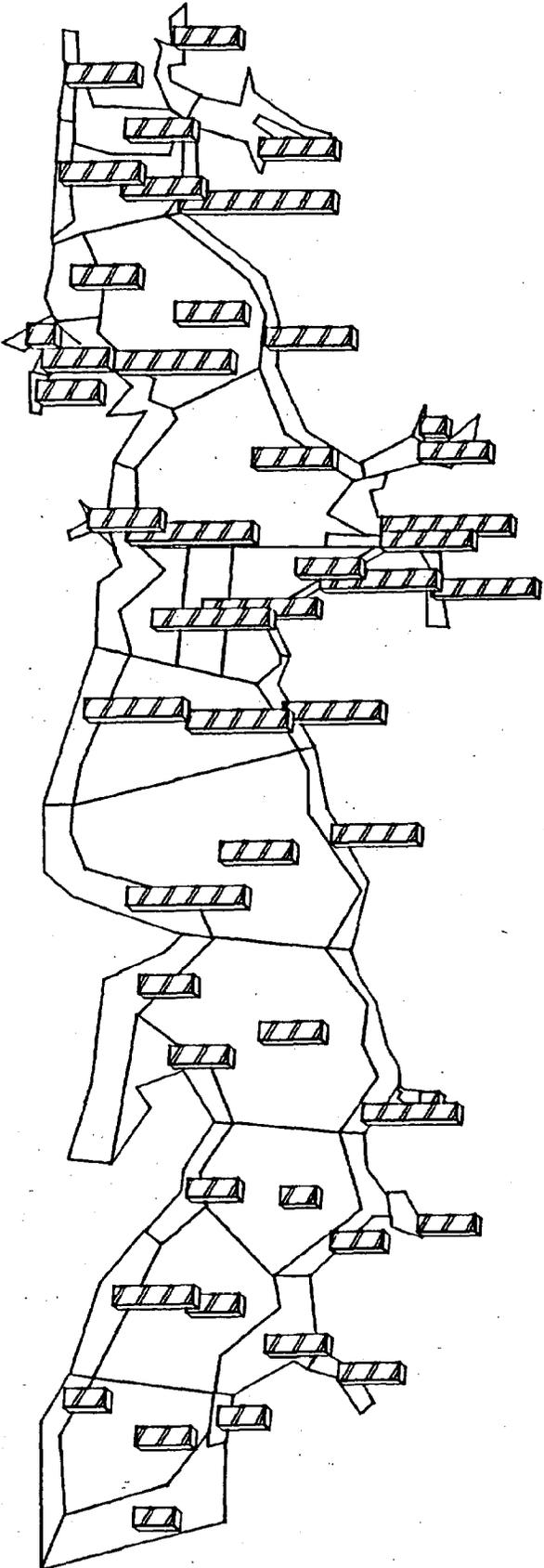
Average of Surface Samples For All Dates



0 to .1 by .001

CHOCTAWHATCHEE BAY

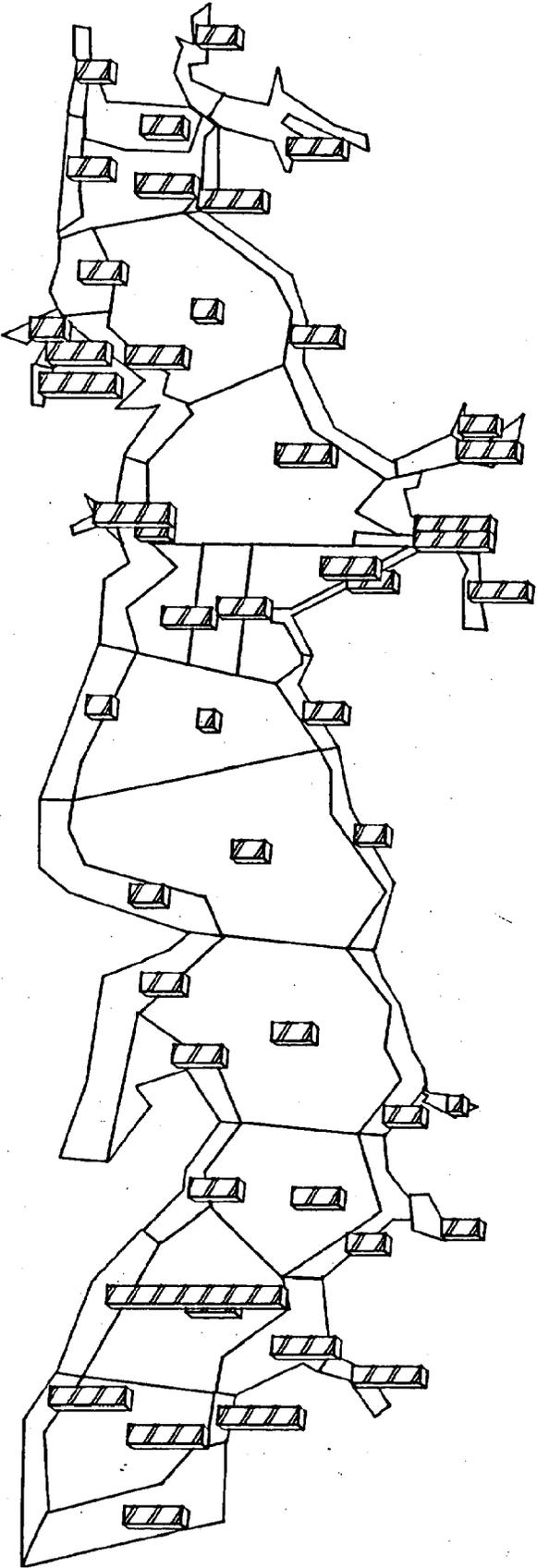
Ammonia in Milligrams Per Liter
Average of Bottom Samples For All Dates



0 to .1 by .001

CHOCTAWHATCHEE BAY

Nitrite in Milligrams Per Liter
Average of Surface Samples For All Dates

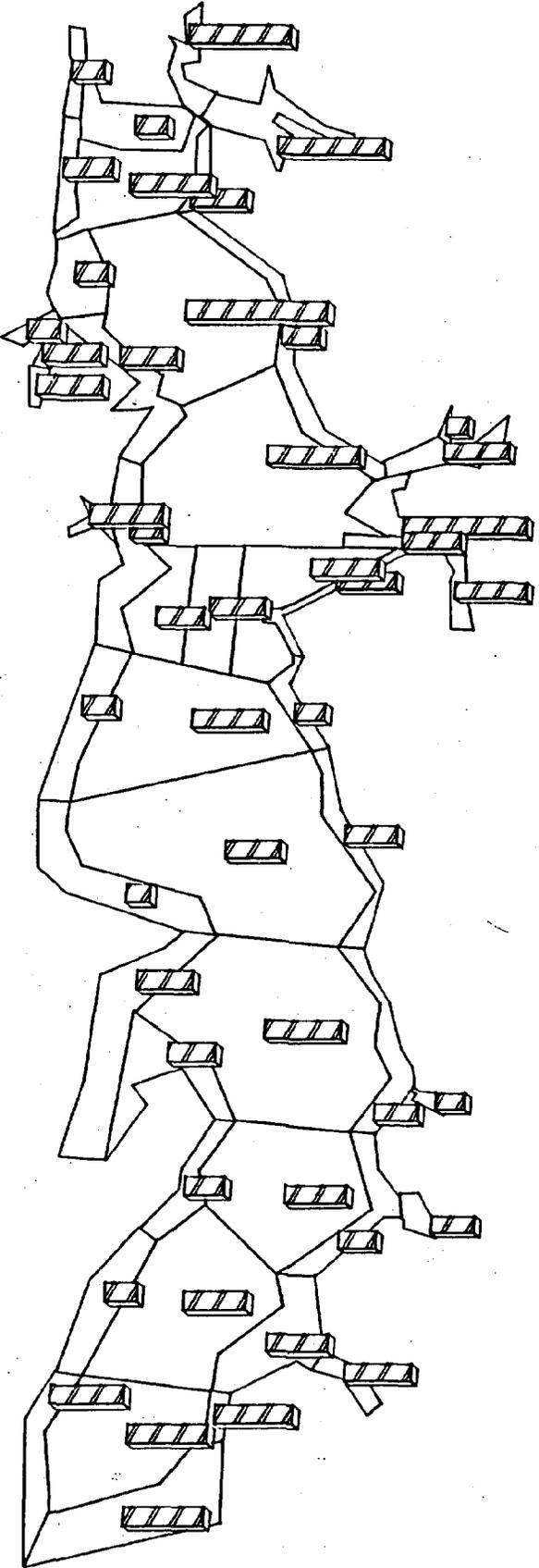


0 to .02 by .001
highest value should be .009
@ sta 8

CHOCTAWHATCHEE BAY

Nitrite in Milligrams Per Liter

Average of Bottom Samples For All Dates

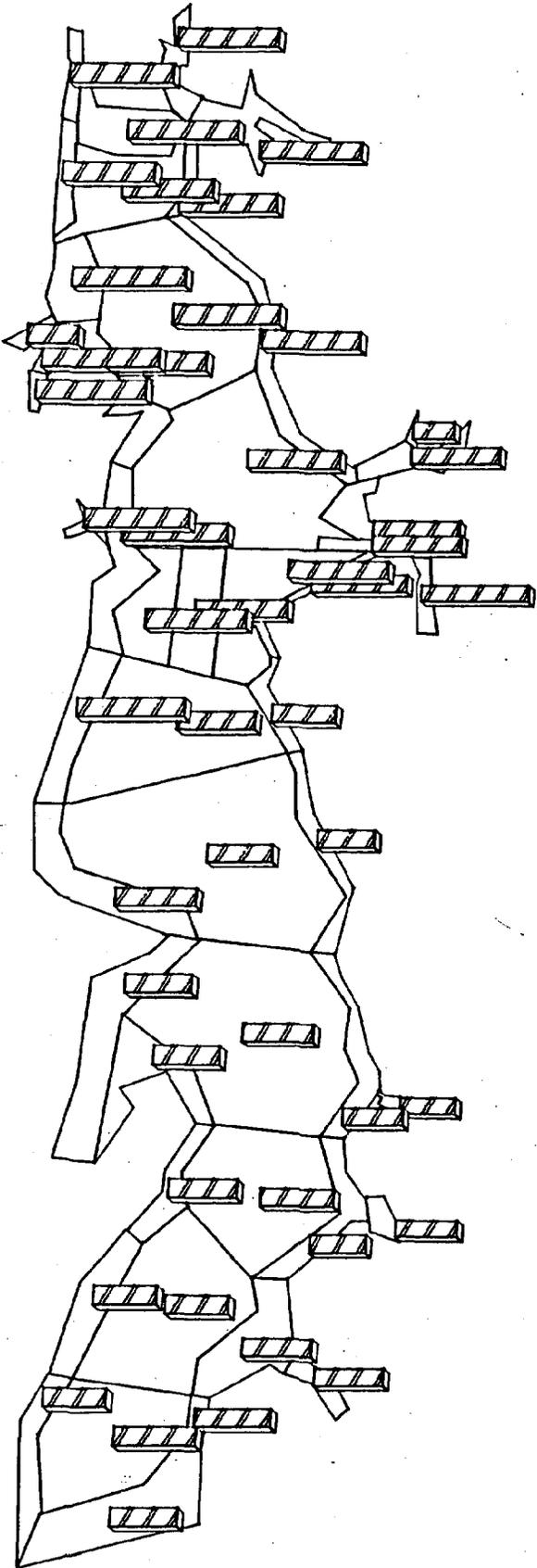


0 to .02 by .001

CHOCTAWHATCHEE BAY

Nitrate in Milligrams Per Liter

Average of Surface Samples For All Dates

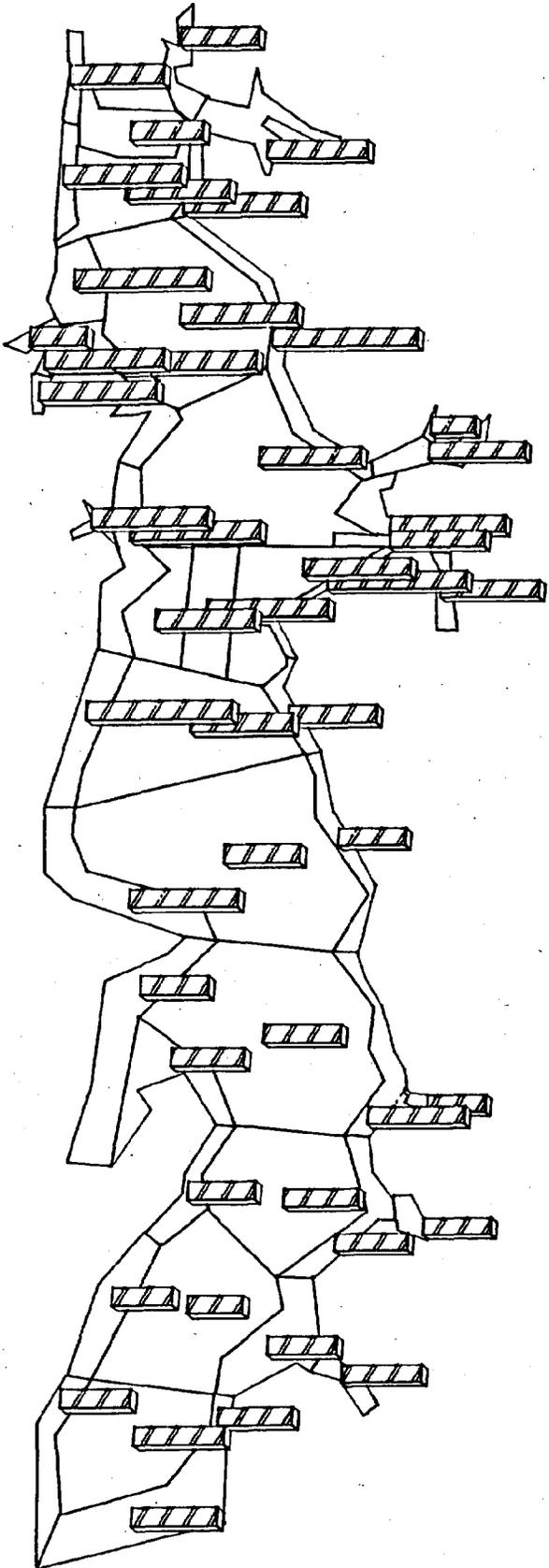


0 to .11 by .001

CHOCTAWHATCHEE BAY

Nitrate in Milligrams Per Liter

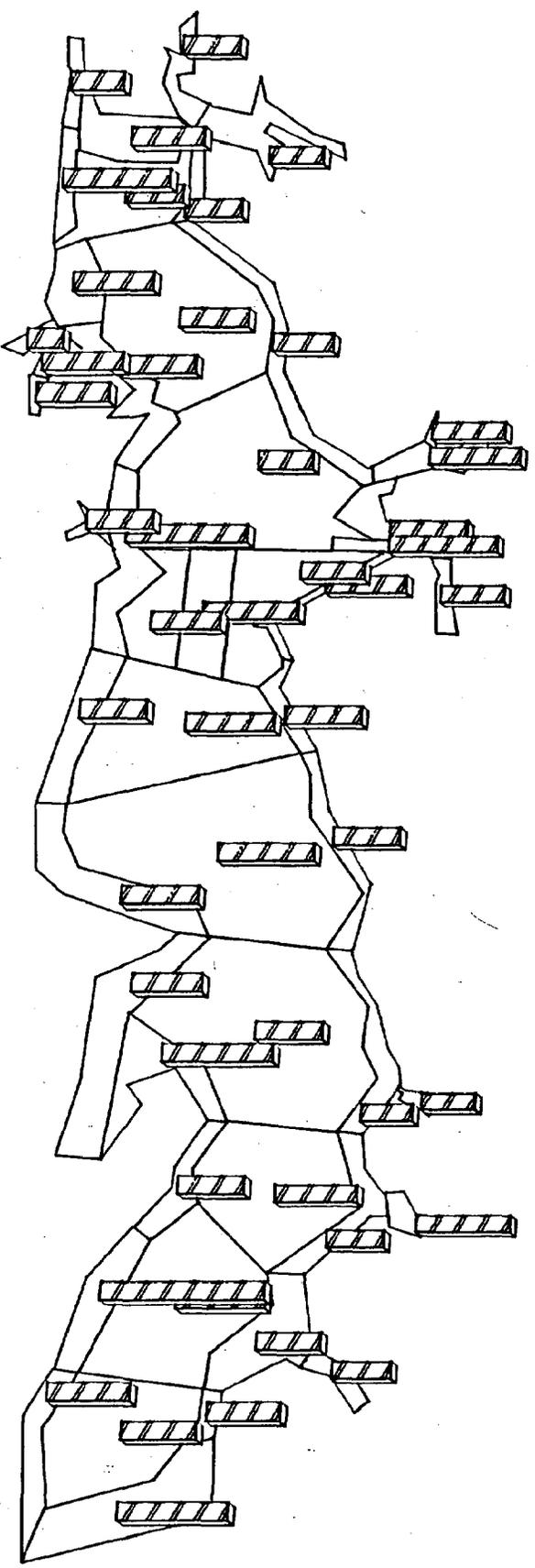
Average of Bottom Samples For All Dates



0 to .11 by .001

CHOCTAWHATCHEE BAY

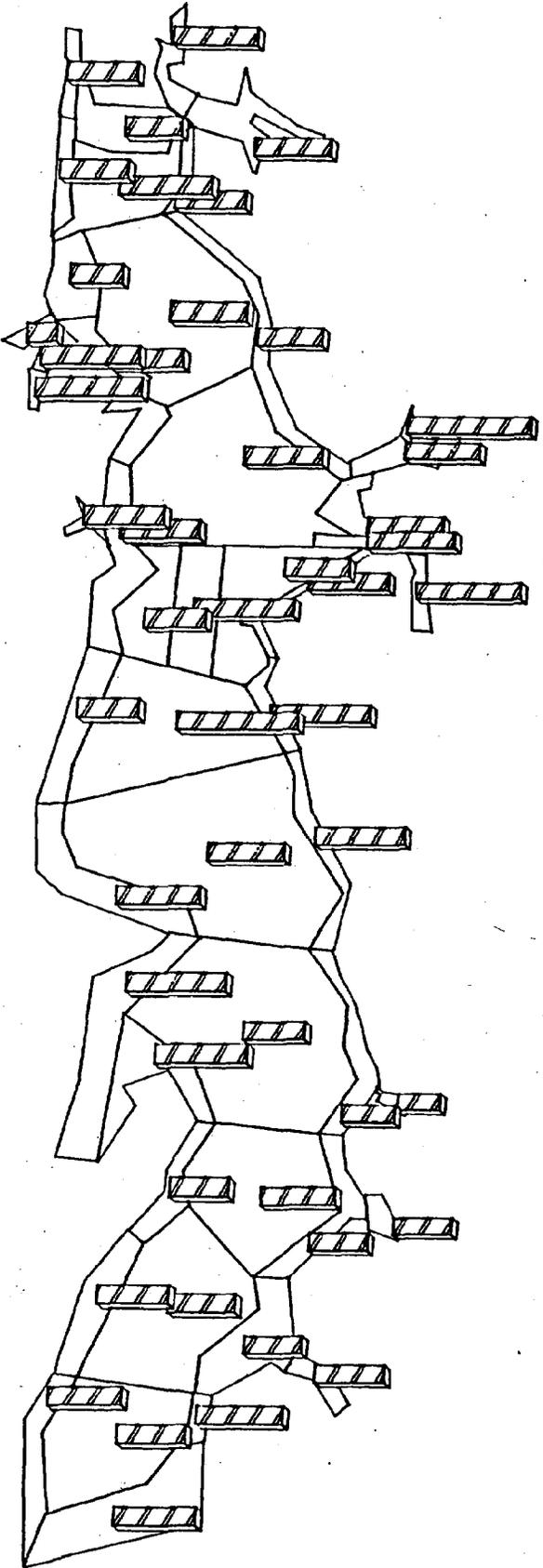
Total Kjeldahl Nitrogen in Milligrams Per Liter
Average of Surface Samples For All Dates



0 to 1.11 by .01

CHOCTAWHATCHEE BAY

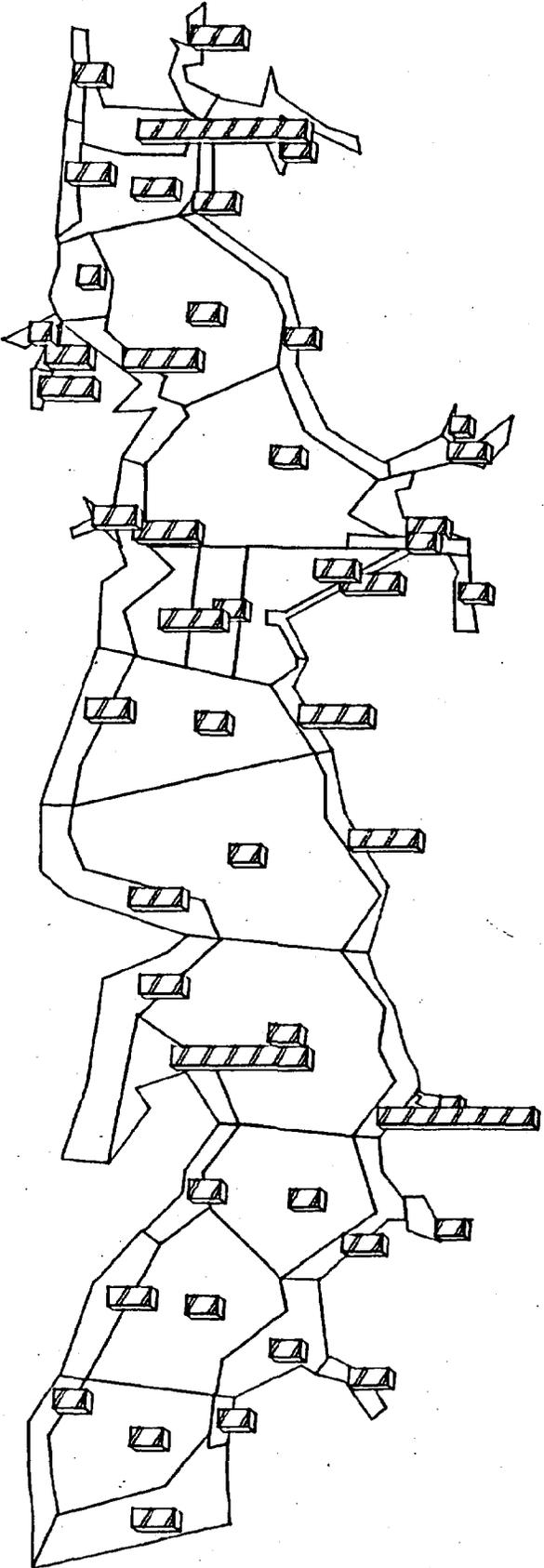
Total Nitrogen in Milligrams Per Liter
Average of Bottom Samples For All Dates



0 to 1.2 by .01

CHOCTAWHATCHEE BAY

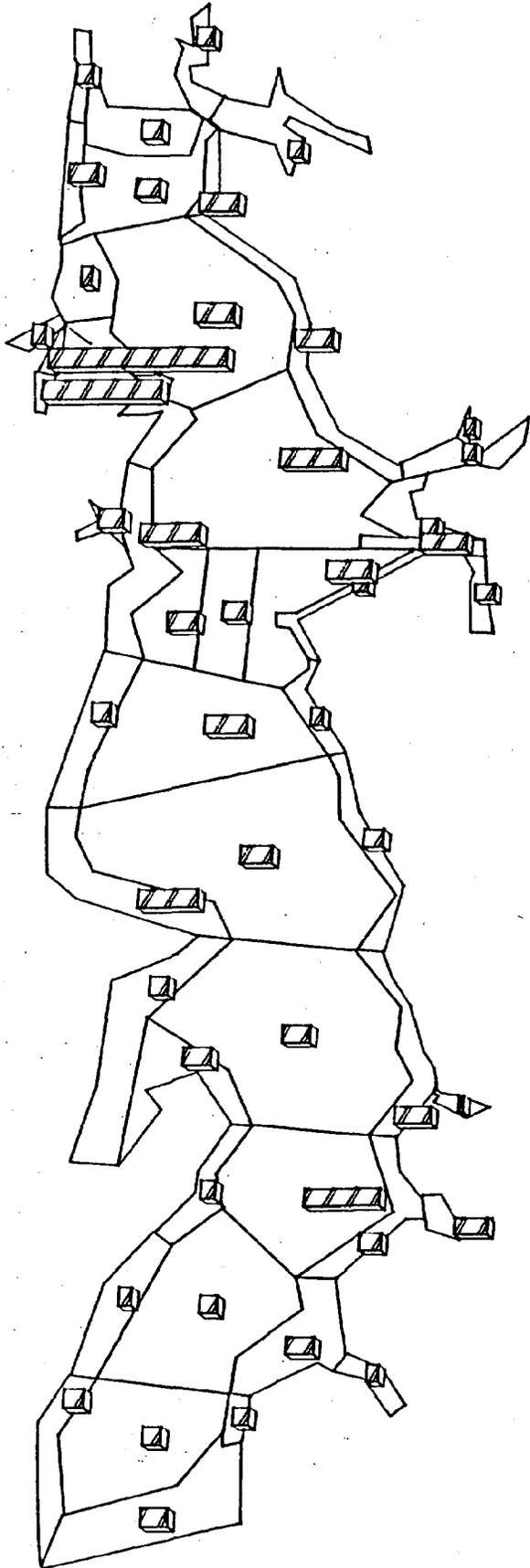
Ortho-phosphate in Milligrams Per Liter
Average of Surface Samples For All Dates



0 to .015 by .001

CHOCTAWHATCHEE BAY

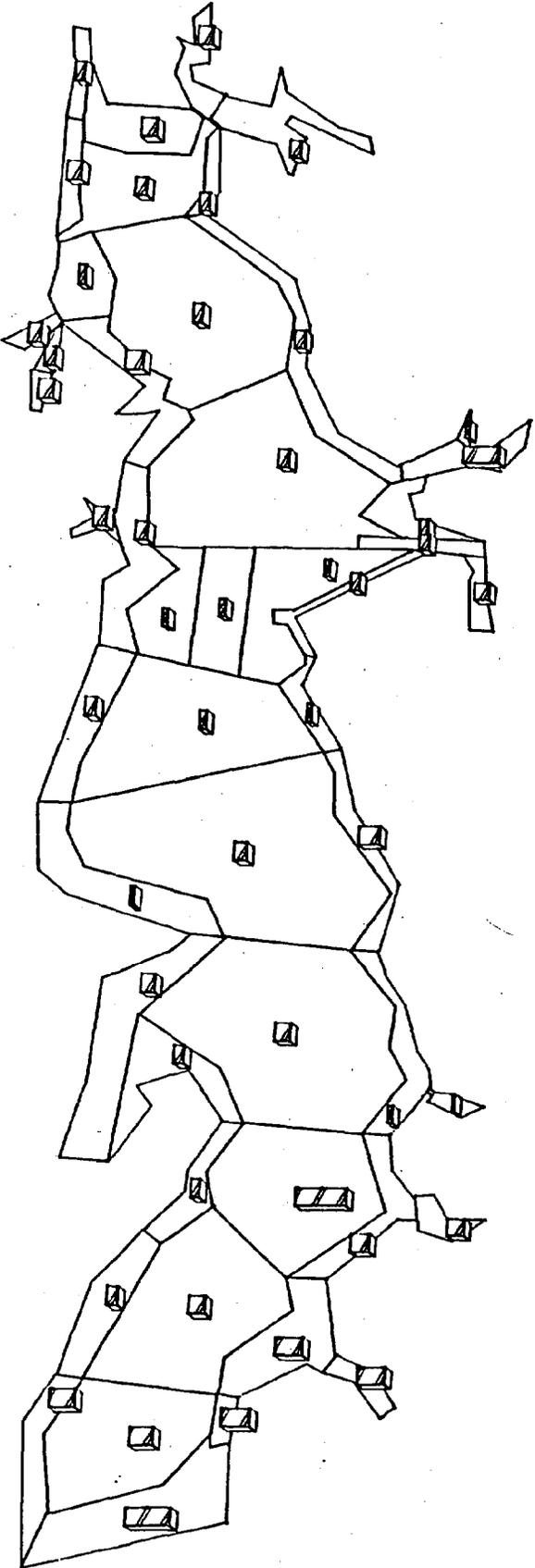
Total Phosphate in Milligrams Per Liter
Average of Bottom Samples For All Dates



0 to .095 by .001

CHOCTAWHATCHEE BAY

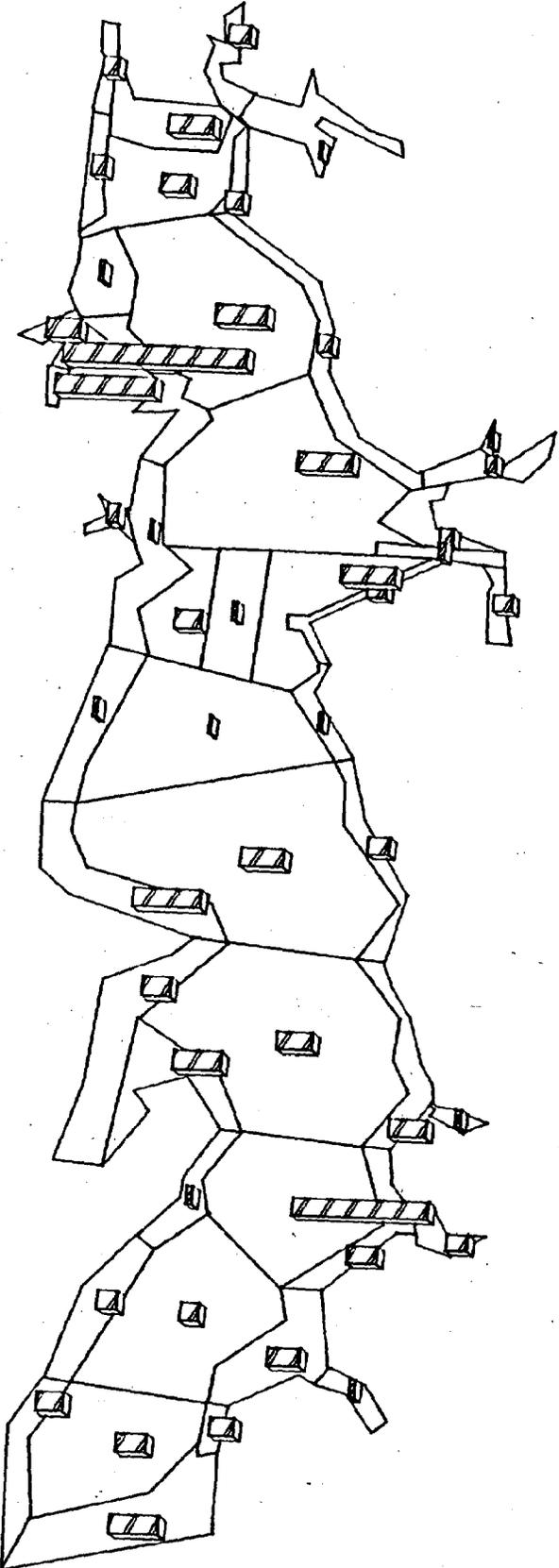
Ratio of Total Phosphate to Total Nitrogen
Average of Surface Samples For All Dates



0 to .2 by .001

CHOCTAWHATCHEE BAY

Ratio of Total Phosphate to Total Nitrogen
Average of Bottom Samples For All Dates



0 to .2 by .001

low dissolved oxygen at various seasons of the year. Areas of particularly low D. O. included lower Rocky Bayou, Boggy Bayou, Tom's Bayou, Garnier Bayou, Cinco Bayou, and Old Pass Lagoon.

Turbidity and color followed similar east-west gradients with the highest levels in areas proximal to the entry point of the Choctawhatchee River. Relatively high color was also noted in some of the northern bayous during the period from December through March. Such factors were inversely related to Secchi depth readings. Overall, the vertical stratification of the Choctawhatchee estuary follows seasonal changes of temperature and salinity. In the late summer, hypoxic conditions are widespread throughout the bay at depth which is an important water quality condition in the system.

Ammonia levels were highest at depth in mid-sections and western portions of the bay. Such concentrations were highest during September and October. Nitrite-N and total N levels were lowest in mid-portions and northeastern sections of the estuary. Nitrate-N was highest in western areas. Peak nitrogen levels occurred during winter months. Orthophosphate was highest in peripheral stations in eastern and western sections of the bay. Mean bottom orthophosphate was high in Old Pass Lagoon (station 36E), peaking during fall months. Average total phosphorous was particularly high in Old Pass Lagoon where the P/N ratios were also high. This area of bay has been described in detail by Livingston (1986b). This area is poorly flushed and receives anthropogenous contamination from urban areas and a marina. Nitrogen and phosphorous nutrients and particulate organic matter (POM) were higher in Old Pass Lagoon than in other portions of the bay and such differences were shown in an ordering of the data (Table 2). Cultural eutrophication was evident in the increased numbers of phytoplankton in Old Pass Lagoon. This basin was also hypoxic at depth, especially in eastern

surface and bottom station data pooled).

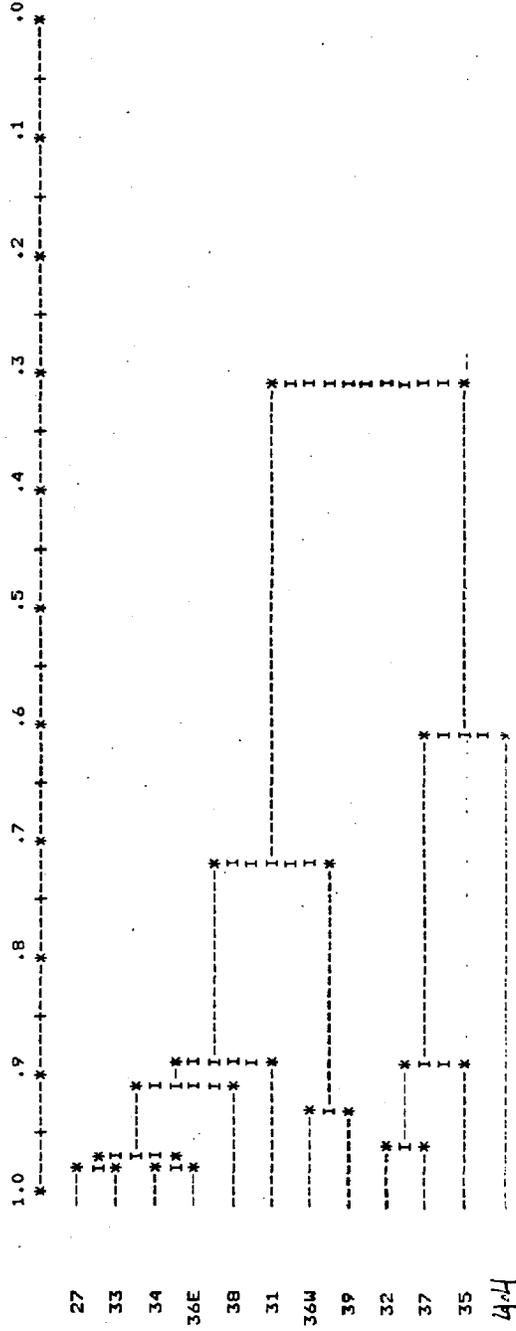
NO2,NO3,TKN CLUSTER - ALL DATES, TOP & BOTTOM AVERAGED

CLUSTERING STRATEGY IS FLEXIBLE GROUPING (WITH BETA)
SIMILARITY COEFFICIENT IS CZEKANOWSKI

CLUSTER LEVEL JOINS NAME WITH SUBGROUP (WHERE GROUP NAME NOW REFERS TO A CLUSTER CONTAINING THE FOLLOWING CLUSTER UNITS)

CLUSTER LEVEL	GROUP NAME	WITH SUBGROUP	27	33	34	36E	38	39	36W	35	36E	38	39
.9810	27	33	27	33									
.9808	34	36E	34	36E									
.9656	27	34	27	33	34	36E							
.9584	32	37	32	37									
.9253	36W	39	36W	39									
.9057	27	38	27	33	34	36E	38						
.8926	27	31	27	31	33	34	36E	38					
.8851	32	35	32	35	37								
.7177	27	36W	27	31	33	34	36E	38	36W	35	36E	38	39
.6072	32	44	32	35	37	44							
.3084	27	32	27	31	32	33	34	35	36W	35	36E	38	39

NO2,NO3,TKN CLUSTER - ALL DATES, TOP & BOTTOM AVERAGED
DENDROGRAM OUTPUT
MINIMUM DISTANCE = .3084



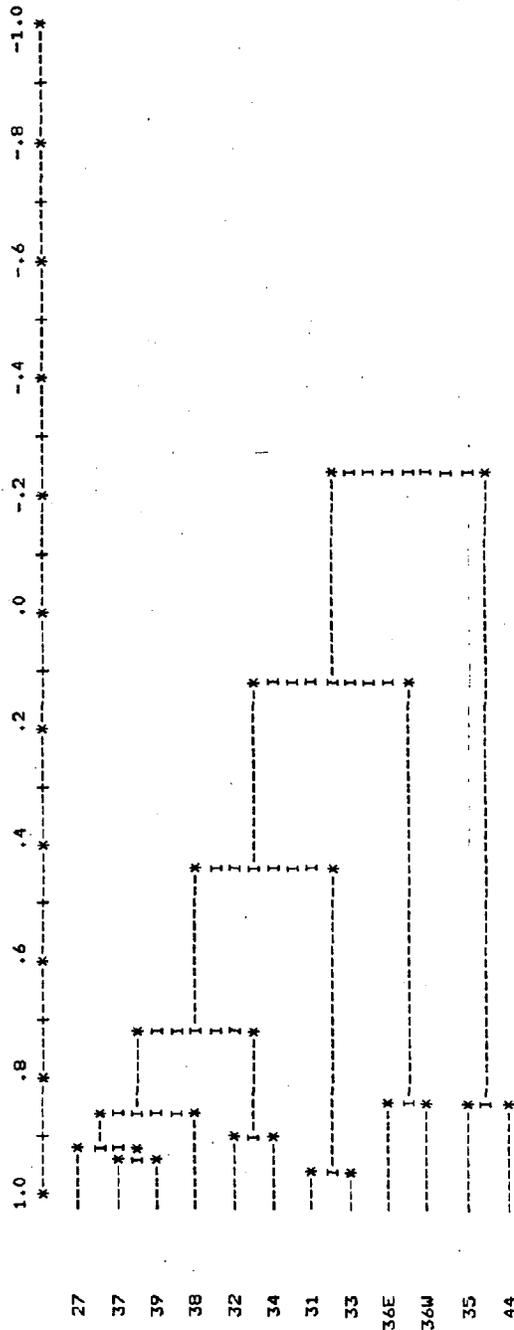
ORTHO AND TOTAL PHOSPHATE - ALL DATES, T & B POOLED

CLUSTERING STRATEGY IS FLEXIBLE GROUPING (WITH BETA)
SIMILARITY COEFFICIENT IS CZEKANOWSKI

CLUSTER LEVEL JOINS GROUP WITH SUBGROUP (WHERE GROUP NAME NOW REFERS TO A CLUSTER CONTAINING THE FOLLOWING CLUSTER UNITS)

CLUSTER LEVEL	GROUP NAME	WITH SUBGROUP	(WHERE GROUP NAME NOW REFERS TO A CLUSTER CONTAINING THE FOLLOWING CLUSTER UNITS)
.9640	31	33	31 33
.9305	37	39	37 39
.9128	27	37	27 37 39
.9026	32	34	32 34
.8550	27	38	27 37 38 39
.8496	35	44	35 44
.8370	36E	36W	36E 36W
.7134	27	32	27 32 34 37 38 39
.4357	27	31	27 31 32 33 34 37 38 39
.1119	27	36E	27 31 32 33 34 36E 37 38 39
-.2457	27	35	27 31 32 33 34 35 36E 37 38 39 44 (ALL ONE GROUP)

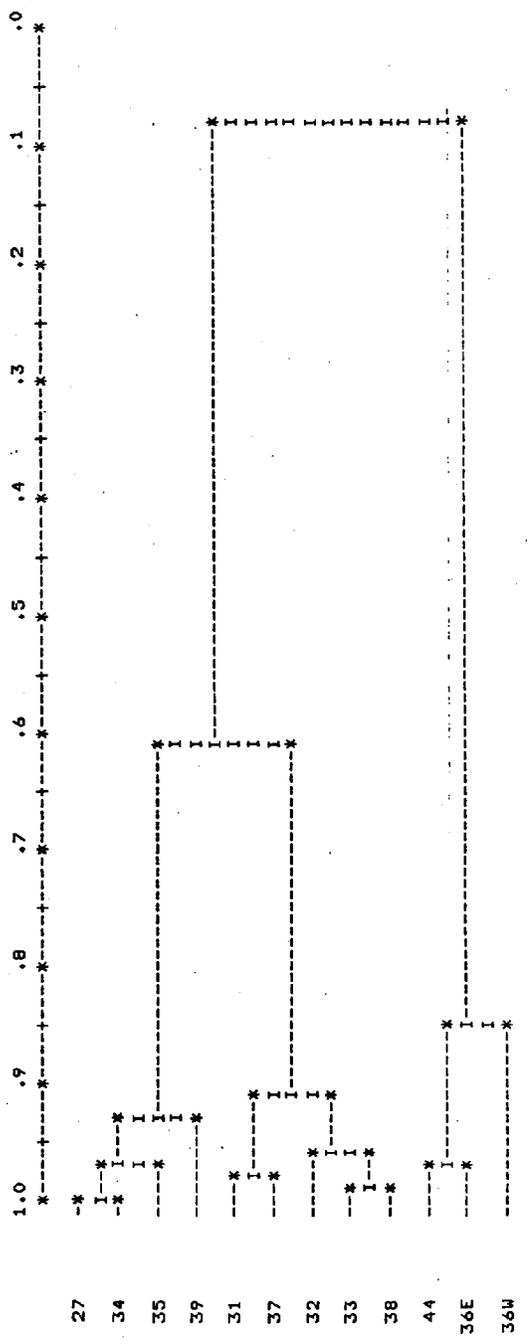
ORTHO AND TOTAL PHOSPHATE - ALL DATES, T & B POOLED
DENDROGRAM OUTPUT
MINIMUM DISTANCE = -.2457



CLUSTER GROUP WITH SUBGROUP (WHERE GROUP NAME NOW REFERS TO A CLUSTER CONTAINING THE FOLLOWING CLUSTER UNITS)

CLUSTER LEVEL	GROUP NAME	WITH SUBGROUP	(WHERE GROUP NAME NOW REFERS TO A CLUSTER CONTAINING THE FOLLOWING CLUSTER UNITS)
.9975	27	34	27 34
.9945	33	38	33 38
.9848	31	37	31 37
.9714	27	35	27 34 35
.9686	44	36E	44 36E
.9550	32	33	32 33 38
.9281	27	39	27 34 35 39
.9141	31	32	31 32 33 37 38
.8524	44	36W	44 36E 36W
.6134	27	31	27 31 32 33 34 35 37 38
.0760	27	44	27 31 32 33 34 35 36E 36W 37 38

PARTICULATE ORGANIC MATTER (SEP 85 - FEB 86) SURFACE & BOTTOM AVERAGED
 DENDROGRAM OUTPUT
 MINIMUM DISTANCE = .0760



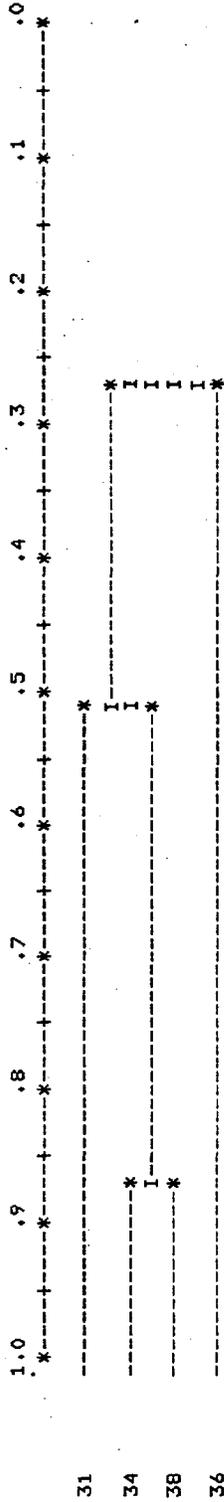
SURFACE PHYTOPLANKTON COUNTS (SEP 85 - JAN 86)

CLUSTERING STRATEGY IS FLEXIBLE GROUPING (WITH BETA)
SIMILARITY COEFFICIENT IS CZEKANOWSKI

CLUSTER LEVEL JOINS NAME WITH SUBGROUP (WHERE GROUP NAME NOW REFERS TO A CLUSTER CONTAINING THE FOLLOWING CLUSTER UNITS)

CLUSTER LEVEL	JOINS NAME	WITH SUBGROUP	(WHERE GROUP NAME NOW REFERS TO A CLUSTER CONTAINING THE FOLLOWING CLUSTER UNITS)
.8723	34	38	34 38
.5057	31	34	31 34 38
.2719	31	36	31 34 36 38 (ALL ONE GROUP)

SURFACE PHYTOPLANKTON COUNTS (SEP 85 - JAN 86)
DENDROGRAM OUTPUT
MINIMUM DISTANCE = .2719



sections of the lagoon. According to Livingston (1987), water and sediments in the Choctawhatchee estuary were relatively clear of a broad spectrum of organic compounds such as organochlorine pesticides, polychlorinated biphenyls, dioxin, organophosphorous pesticides, and chlorinated herbicides. Eastern and central portions of the estuary had relatively high metal concentrations in the sediments which also had high silt/clay fractions. High metal concentrations were also found in the urbanized bayous in the western section of the bay (Boggy, lower Rocky, Garnier, Old Pass Lagoon) with stormwater runoff and marinas as the most likely sources of pollutants. Infaunal macroinvertebrate associations indicated degraded conditions in Old Pass Lagoon which had high levels ("enriched" as defined by FDER guidelines) of cadmium, copper, lead, and zinc in the sediments (Table 3).

Chlorophyll a (Figure 3) was found in the highest concentrations in eastern portions of the bay. Particulate organic matter was highest in Old Pass Lagoon and some of the eastern bay stations (Figure 4).

2. Taxonomic review: species occurrence

The highest overall numbers of phytoplankton were noted in Old Pass Lagoon and the westernmost sampling station (Figure 5). These stations were also highest in phytoplankton species richness. Such data indicate that the nutrient-enriched waters of the western bay stations that receive stormwater runoff from urbanized areas and/or are the sites for major marinas are subject to major increases in phytoplankton numbers. However, such high concentrations could also be due, in part, to reduced predation due to low numbers of zooplankton in addition to the more obvious correlation with high nutrient levels (Figure 6). Overall, the general distribution of phytoplankton in the bay indicates

Table 3: Metal accumulation index (A) computed for seven metals at 25 locations sampled in sediments of Choctawhatchee Bay in April, 1987.

TA	Arsenic	Cadmium	Copper	Chromium	Lead	Nickel	Zinc
01	20.56 ¹	0.93	1.59	0.74	1.32	0.83	1.27
03	5.00 ²	1.60	4.47 ²	2.74	1.42	2.40 ²	1.81
05	4.87 ¹	2.48	5.82 ¹	0.77	2.09 ¹	1.43	1.61
07	2.00	1.42	5.04 ²	2.87	1.22	2.67 ²	1.96
09	3.69	0.93	0.92	0.78	2.03 ¹	0.38	0.89
11	5.21 ²	1.00	5.03 ²	2.77	1.32	2.50 ²	1.81
13	6.22 ²	3.27	2.64	3.20 ²	2.69 ²	2.03	1.67
15	3.00	1.10	1.63	1.88	1.35	1.35	1.26
17	5.24 ¹	0.36	2.18	2.32	1.94	1.83	1.33
19	7.25 ²	1.69	7.64 ²	3.63 ²	1.77	4.65 ²	2.42 ²
20	1.62	0.38	0.57	0.92	1.56	0.76	0.78
25	5.41 ²	1.01	2.74	2.35	1.70	2.39 ²	1.60
6B	4.91 ¹	2.41	3.04 ¹	1.16	2.22 ¹	2.34 ¹	1.89
27	0.57	1.02	1.29	0.87	2.54 ¹	0.93	1.25
28	1.28	2.21	2.91 ¹	2.16	2.61 ¹	2.58 ¹	1.70
29	1.82	4.81 ¹	3.70 ¹	3.79 ¹	4.57 ¹	2.94 ¹	2.35 ¹
30	3.45	10.35 ¹	1.90	1.56	2.50 ¹	0.97	1.59
31	5.77 ¹	1.55	5.75 ¹	0.92	2.44 ¹	2.39 ¹	1.71
32	0.90	1.58	1.12	0.90	1.99	0.70	1.21
34	3.21	3.05	2.33	2.02	2.23 ¹	2.19	1.73
6E	2.37	5.21 ¹	13.36 ¹	1.93	9.29 ¹	1.55	11.17 ¹
37	18.83 ¹	0.94	0.97	0.88	6.01 ¹	0.77	1.01
38	3.26	0.34	0.48	0.49	3.21 ¹	0.78	1.02
40	1.99	4.88 ¹	3.92 ¹	2.96	5.91 ¹	2.82 ¹	3.04 ¹
42	0.84	0.91	1.70	0.65	3.74 ¹	0.70	2.21

metal enriched at this station

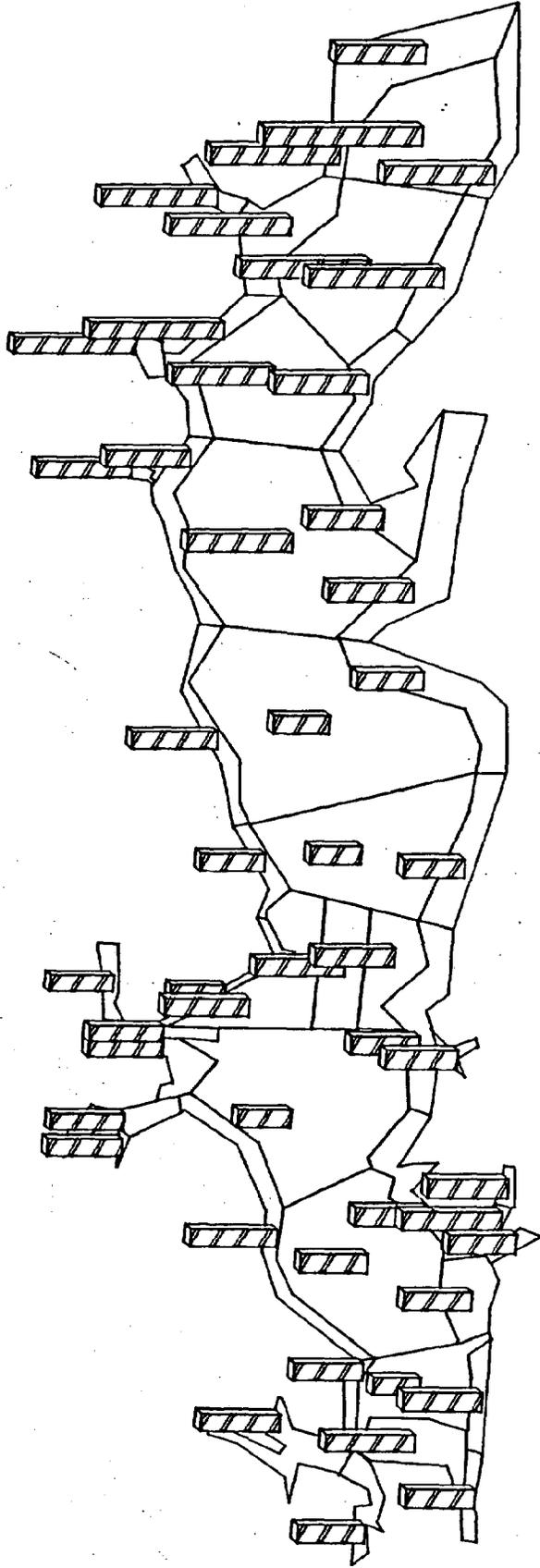
metal possibly enriched at this station (aluminum concentration > 79,000 ppm precludes assignment as enriched)

through August, 1966.

CHOCTAWHATCHEE BAY

Chlorophyll a in Milligrams Per Cubic Meter

Average of Surface Samples For All Dates

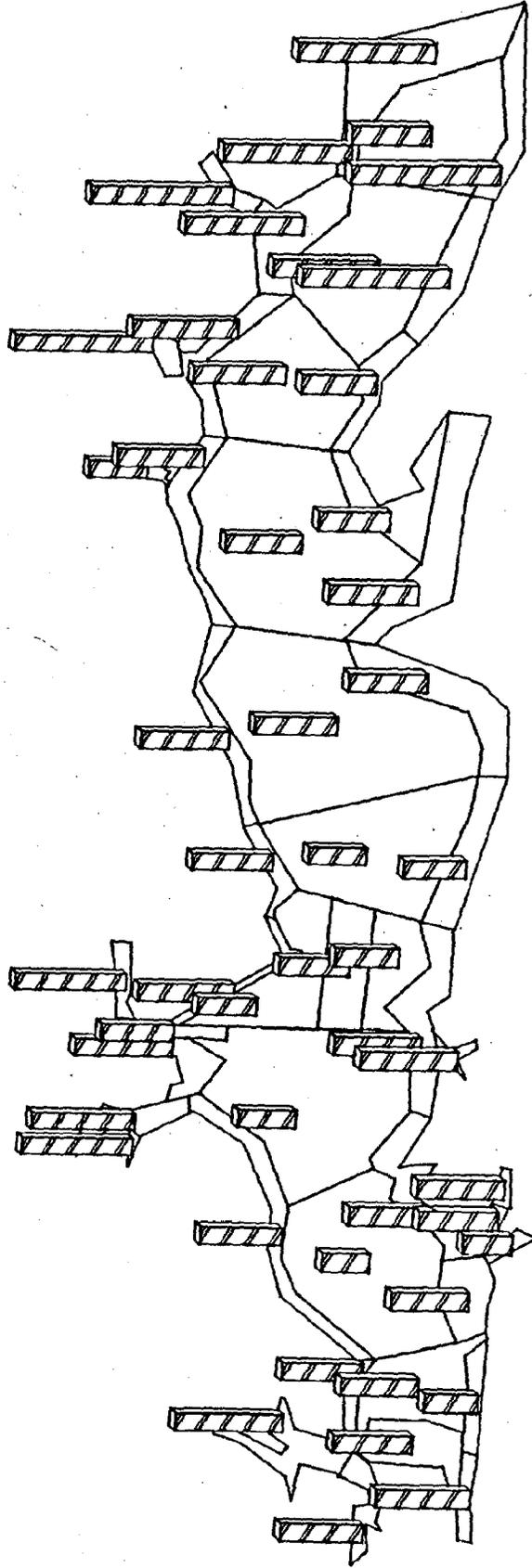


0 to 3.63 by .01

CHOCTAWHATCHEE BAY

Chlorophyll a in Milligrams Per Cubic Meter

Average of Bottom Samples For All Dates

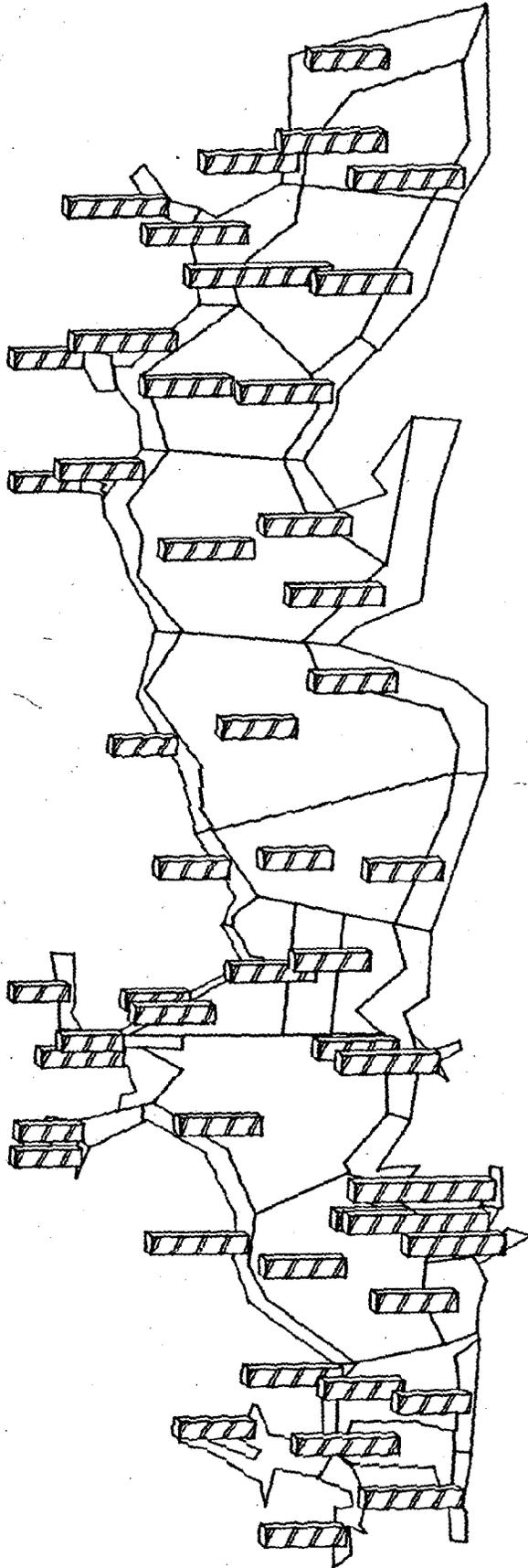


0 to 3.63 by .01

September, 1985 through August, 1986.

CHOCTAWHATCHEE BAY

Particulate Organic Matter in Milligrams Per Liter
Average of Surface Samples For All Dates

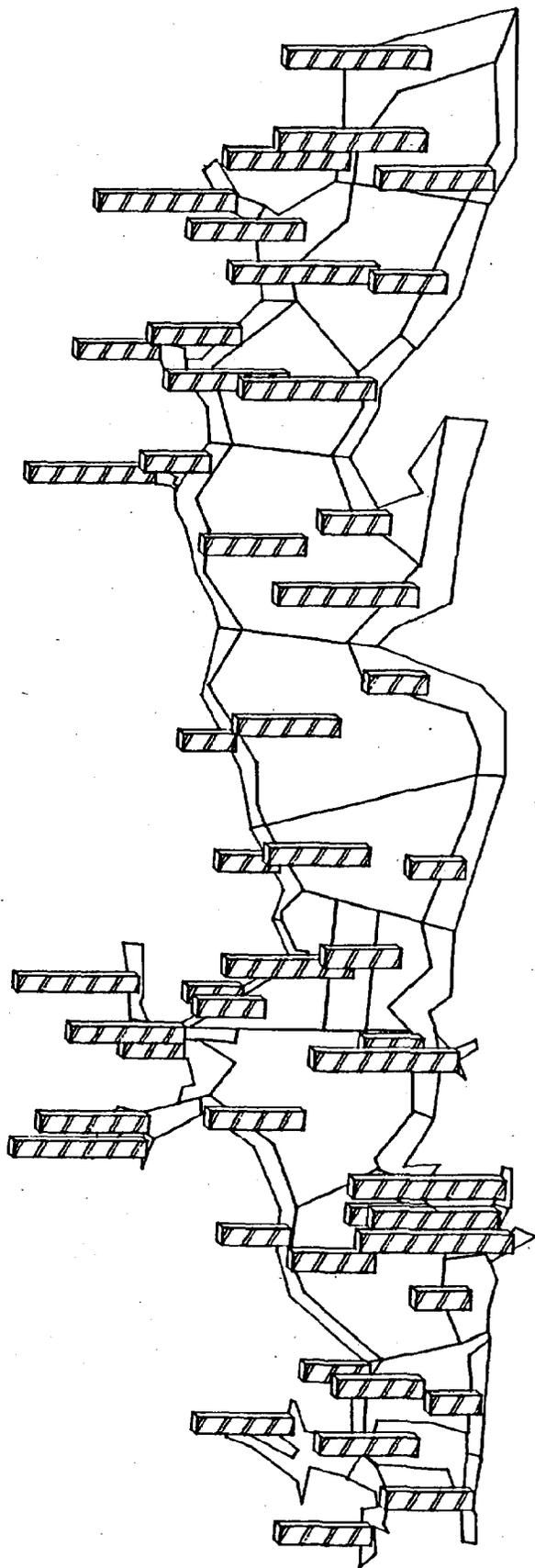


© to 5.9 by .1

CHOCTAWHATCHEE BAY

Particulate Organic Matter in Milligrams Per Liter

Average of Bottom Samples For All Dates

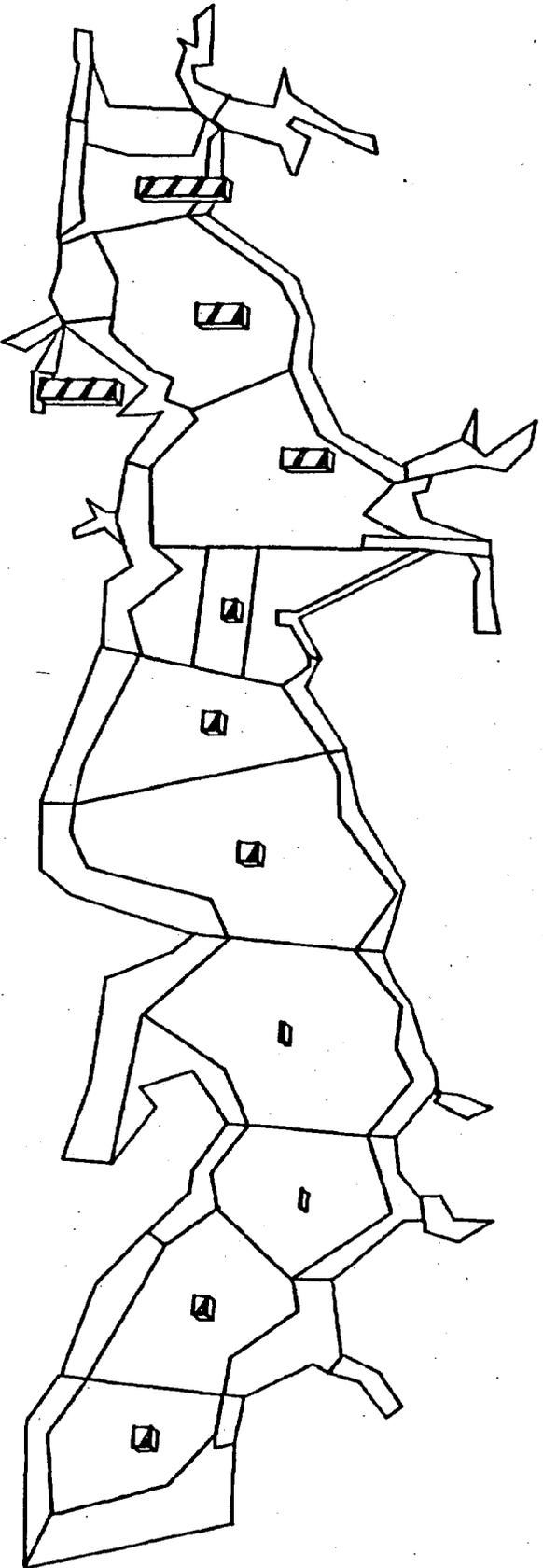


0 to 5.9 by .1

Figure 5: Summary data (12 month averages) of phytoplankton numbers and species richness taken during the day in the Choctawhatchee Bay system from September, 1985 through August, 1986.

CHOCTAWHATCHEE BAY

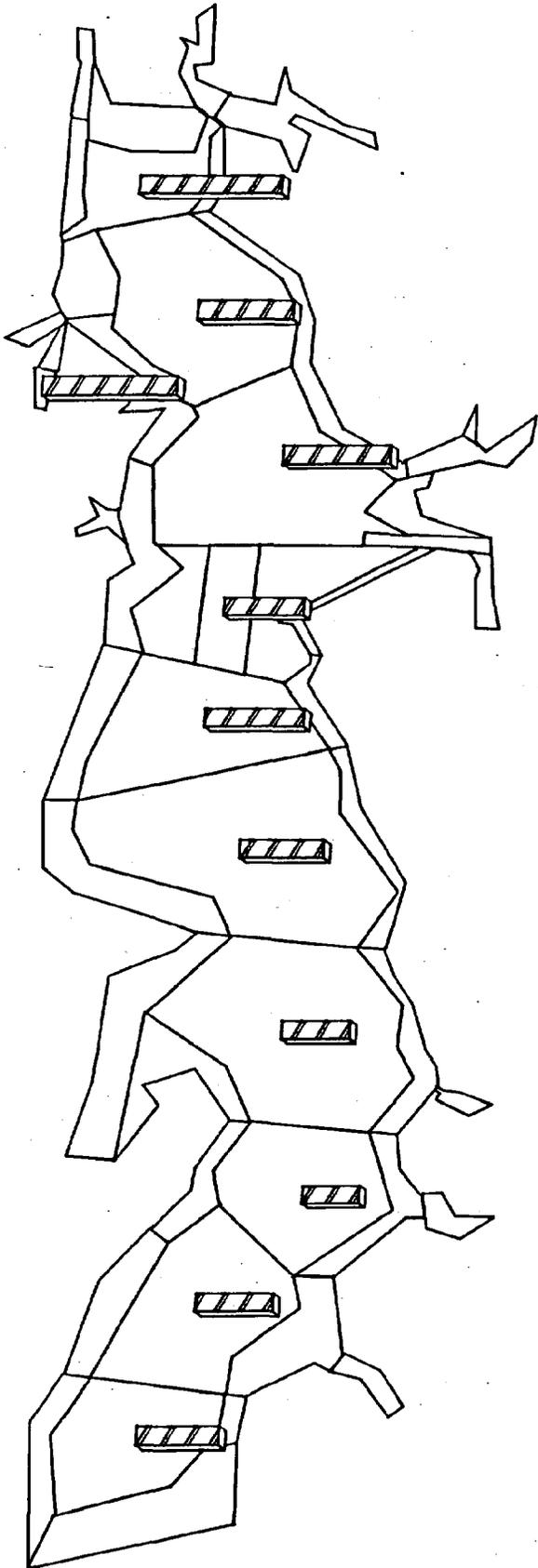
Phytoplankton in Number of Individuals Per Cubic Meter
Average of Day Surface Samples For All Dates



2,200,000 to 24,000,000
by 100,000

CHOCTAWHATCHEE BAY

Phytoplankton in Number of Species
Average of Day Surface Samples For All Dates

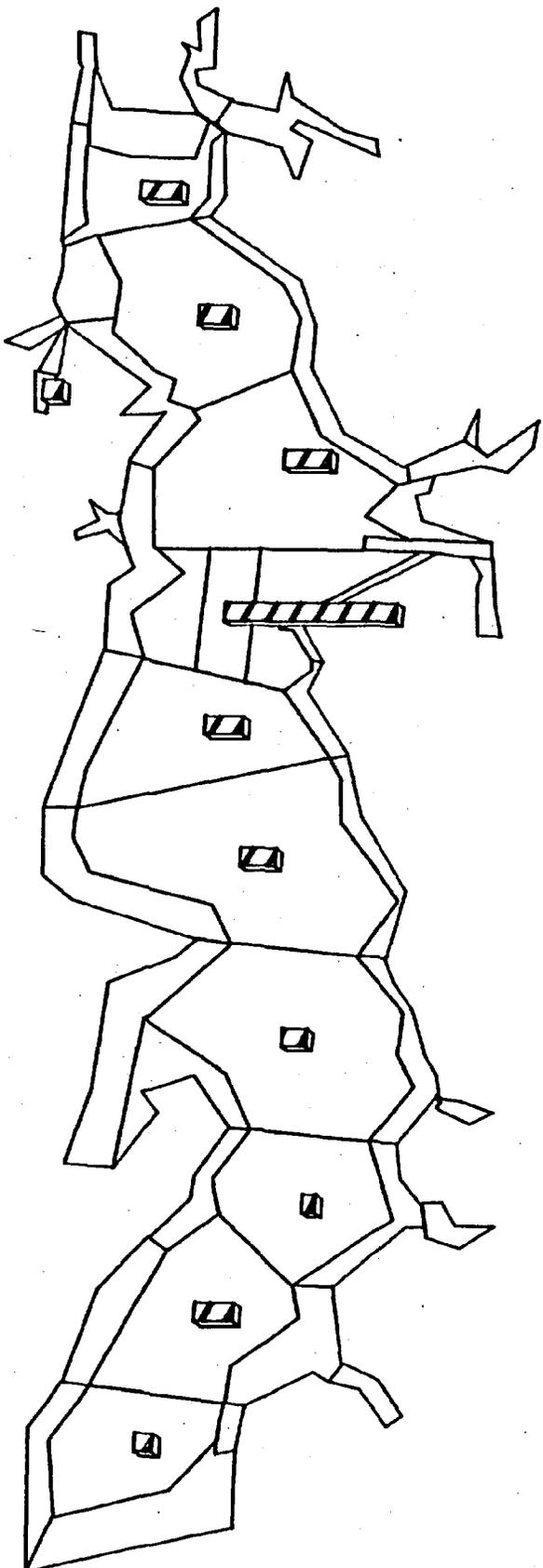


0 to 46 by 1

Figure 6: Summary data (12 month averages) of physical-chemical taken in the Choctawhatchee Bay system from September, 1985 through August, 1986.

CHOCTAWHATCHEE BAY

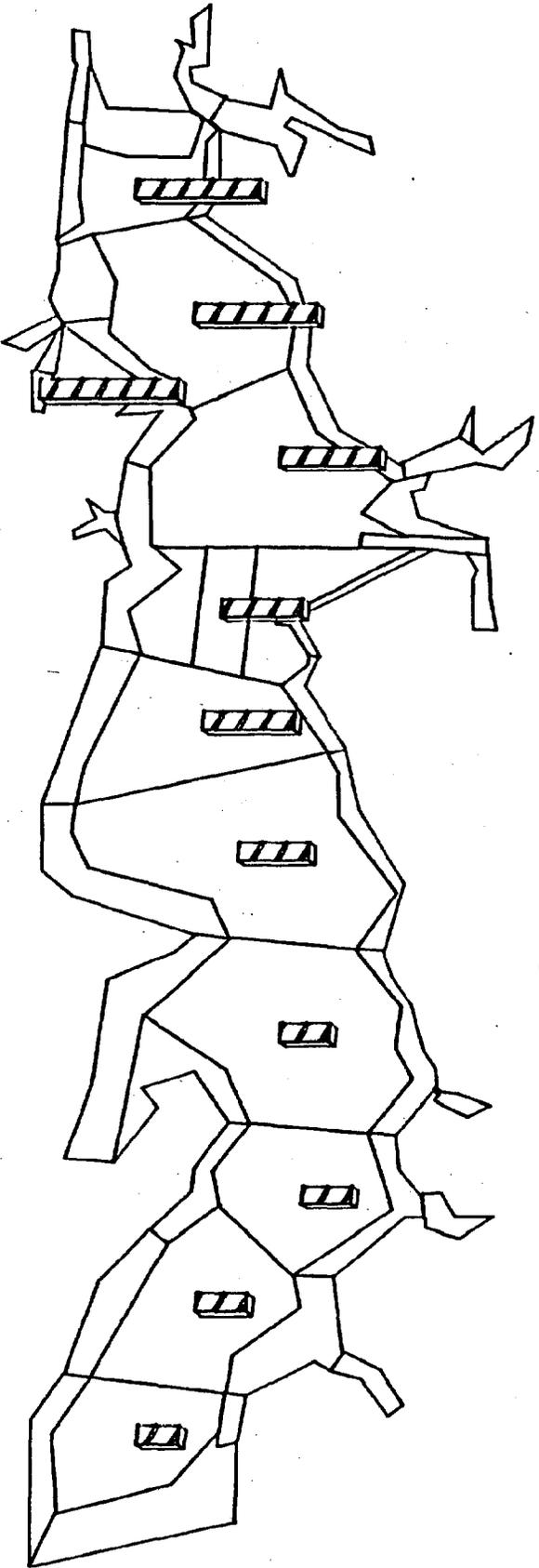
Zooplankton in Number of Individuals Per Cubic Meter
Average of Day Surface Samples For All Dates



CHOCTAWHATCHEE BAY

Zooplankton in Number of Species
Average of Day Surface Samples For All Dates

0 to 16 by 1



a positive (direct) relationship with areas enriched by chemicals associated with anthropogenous activities.

A review of the spatial/temporal occurrence frequency of estuarine algae in Choctawhatchee Bay is given in Table 4. Two genera were dominant in terms of the numbers of species: *Navicula* and *Nitzschia*. In terms of the most frequently taken species, there was a series of dominants: *Ceratium hircus*, *Cyclotella striata*, *Cyclotella* sp., *Chaetoceros decipiens*, *C. radicans*, *C. brevis*, *Coscinodiscus centralis*, and *C. granii*. Of these species, a group of dominants was chosen for an analysis of distribution along specific habitat gradients. Such species (16) were chosen as frequency dominants (occurring during all times of the year) or as numerical dominants (the top species in terms of numbers taken over the entire sampling period). It can be argued that sub-dominant and rare species can be important indicators of water quality. However, for the purposes of this study, only the dominants were treated as a function of water quality factors that include salinity, dissolved oxygen, and various nutrients.

3. Algae as water quality indicators

Numbers of individuals, numbers of species, and the Shannon diversity index were used as phytoplankton community indices. The data (Figure 7) indicate that the station in the western section of the bay that was characterized by high nutrient levels (36E) was also characterized by periodic peaks of phytoplankton numbers (in this case, the late fall and early winter months). Relatively high numbers of phytoplankton were also found at station 38 which was subject to stormwater runoff from urbanized areas of the western portions of the bay. Species richness was also high at the eutrophicated stations. Old pass lagoon had relatively high species richness during most of the year with low

Table 4: Systematic review of phytoplankton taken with 25 and 64 μ m nets in the Choctawhatchee estuary from September, 1985 through August, 1986.

Systematic review

Choctawhatchee Bay, day, surface, 25 μ samples

	No. of occurrences
Division...Cyanophyta	
Class.....Cyanophyceae (blue-green algae or cyanobacteria)	
Order.....Chroococcales	
Family.....Chroococcaceae	
<i>Synechocystis</i> sp.	1
Order.....Nostocales	
Family.....Nostocaceae	
<i>Anabaena</i> sp.	1
Family.....Oscillatoriaceae	
<i>Oscillatoria</i> sp. 3	1
Division...Euglenophyta	
Class.....Euglenophyceae	
Order.....Euglenales	
<i>Phacus</i> sp.	1
Division...Cryptophyta	
Class.....Cryptophyceae	
Order.....Cryptomonadales	
Family.....Cryptomonadaceae	
<i>Cryptomonas</i> sp. 1	3
<i>Cryptomonas</i> sp.	1
Division...Dinophyta	
Class.....Dinophyceae (dinoflagellates)	
Unidentified dinoflagellates	1
Order.....Peridinales	
Family.....Peridiniaceae	
<i>Peridinium oblongum</i>	2
<i>Peridinium</i> sp.	1
Order.....Gonyaulacales	
Family.....Ceratiaceae	
<i>Ceratium fusus</i>	3
<i>Ceratium hircus</i>	19
<i>Ceratium tripos</i>	1
Family.....Gonyaulacaceae	
<i>Gonyaulax</i> sp. 1	5
Order.....Prorocentrales	
Family.....Prorocentraceae	
<i>Prorocentrum gracile</i>	1
<i>Prorocentrum micans</i>	13
<i>Pronoctiluca acuta</i>	1
<i>Prorocentrum pyriformis</i>	1
<i>Prorocentrum</i> sp. 1	1
<i>Prorocentrum triestinum</i>	1
Division...Chrysophyta	
Class.....Chrysophyceae (golden-brown algae)	
Order.....Chrysomonadales	
Family.....Ochromonadaceae	
<i>Dinobryon divergens</i>	1
<i>Dinobryon sertularia</i>	2
Division...Xanthophyta	
Class.....Xanthophyceae (yellow-green algae)	

Order.....	Tribonemales	
Family.....	Tribonemataceae	
	<i>Tribonema affine</i>	3
Division...	Bacillariophyta (diatoms)	
Class.....	Bacillariophyceae	
Order.....	Pennales	
Suborder...	Araphidineae	
Family.....	Diatomaceae	
	<i>Asterionella japonica</i>	2
	<i>Ctenophora pulchella</i>	1
	<i>Delphineis surirella</i>	3
	<i>Falcula hyalina</i>	4
	<i>Opephora martyi</i>	2
	<i>Opephora pacifica</i>	4
	<i>Rhabdonema adriaticum</i>	8
	<i>Synedra acus</i>	5
	<i>Synedra sp.</i>	1
	<i>Synedra ulna</i>	8
	<i>Thalassionema nitzschioides</i>	9
	<i>Thalassiothrix longissima</i>	1
Suborder...	Raphidineae	
Family.....	Eunotiaceae	
	<i>Eunotia alpina</i>	3
	<i>Eunotia pectinalis</i>	5
	<i>Eunotia serra</i>	1
Family.....	Achnantheaceae	
	<i>Cocconeis placentula</i>	7
	<i>Cocconeis pseudomarginata</i>	1
	<i>Cocconeis scutellum</i>	1
Family.....	Naviculaceae	
	<i>Amphiprora gigantea</i>	9
	<i>Amphora coffeaeformis</i>	2
	<i>Amphora commutata</i>	1
	<i>Amphora ovalis</i>	1
	<i>Caloneis acutiascula</i>	2
	<i>Caloneis formosa</i>	2
	<i>Caloneis latiascula (new var cr 249)</i>	1
	<i>Caloneis latiascula</i>	5
	<i>Caloneis permagna</i>	1
	<i>Caloneis westii</i>	1
	<i>Capartogramma crucicula</i>	2
	<i>Cymbella gracilis</i>	1
	<i>Diploneis elliptica</i>	1
	<i>Diploneis ovalis</i>	5
	<i>Diploneis parma</i>	1
	<i>Diploneis sp. 1</i>	1
	<i>Frustulia rhomboides</i>	2
	<i>Gomphonema angustatum</i>	3
	<i>Gomphonema gracile</i>	1
	<i>Gyrosigma acuminatum</i>	1
	<i>Gyrosigma macrum</i>	1
	<i>Gyrosigma spenceri</i>	1
	<i>Navicula cf. ammophila</i>	4
	<i>Navicula bailyana</i>	1
	<i>Navicula distans</i>	2
	<i>Navicula longa</i>	1
	<i>Navicula lyra</i>	6
	<i>Navicula pupula</i>	2
	<i>Navicula salsa</i>	2

<i>Navicula</i> sp. 1	1
<i>Navicula viridula</i>	2
<i>Navicula yarrensis</i> v. <i>americana</i>	1
<i>Navicula yarrensis</i>	14
<i>Neidium iridis</i>	1
<i>Pinnularia gibba</i>	1
<i>Pleurosigma elongatum</i>	4
<i>Pleurosigma</i> sp.	2
<i>Stauroneis phoenicenteron</i>	2
<i>Stauroneis spicula</i>	4
<i>Tropidoneis lepidoptera</i>	2
Family.....Nitzschiaceae	
<i>Bacillaria paxillifer</i>	12
<i>Nitzschia circumscuta</i>	1
<i>Nitzschia acicularis</i> v. <i>closterioides</i>	10
<i>Nitzschia constricta</i>	2
<i>Nitzschia dubia</i>	1
<i>Nitzschia jelinecki</i>	1
<i>Nitzschia lanceolata</i>	1
<i>Nitzschia linearis</i>	2
<i>Nitzschia longissima</i>	9
<i>Nitzschia</i> sp. (needle-like)	1
<i>Nitzschia sigma</i>	3
<i>Nitzschia sigmoidea</i>	6
<i>Nitzschia triblionella</i>	7
Family.....Surirellaceae	
<i>Surirella fastuosa</i>	12
<i>Surirella gemma</i>	5
<i>Surirella ovata</i>	2
<i>Surirella robusta</i>	1
Order.....Centrales	
Suborder...Coscinodiscineae	
Family.....Thalassiosiraceae	
<i>Aulacosira granulata</i>	2
<i>Cyclotella meneghiniana</i>	1
<i>Cyclotella</i> sp. (small)	18
<i>Cyclotella stelligera</i>	1
<i>Cyclotella striata</i>	33
<i>Cyclotella stylorum</i>	7
<i>Skeletonema costatum</i>	7
<i>Thalassiosira decipiens</i>	2
<i>Thalassiosira eccentrica</i>	13
<i>Thalassiosira lineatus</i>	1
<i>Thalassiosira oestrupii</i>	15
Family.....Melosiraceae	
<i>Leptocylindrus danicus</i>	5
<i>Melosira granulata</i>	1
<i>Melosira undulata</i>	2
<i>Melosira varians</i>	3
<i>Paralia sulcata</i>	5
<i>Podosira stelliger</i>	1
Family.....Hemidiscaceae	
<i>Actinocyclus ehrenbergii</i>	1
Family.....Heliopeltaceae	
<i>Actinoptychus undulatus</i>	8
Family.....Eupodiscaceae	
<i>Triceratium arcticum</i>	1
Family.....Chaetoceraceae	
<i>Bacteriastrum varians</i>	1
<i>Chaetoceros atlanticus</i>	4

	<i>Chaetoceros brevis</i>	19
	<i>Chaetoceros compressus</i>	2
	<i>Chaetoceros costatum</i>	1
	<i>Chaetoceros decipiens</i>	18
	<i>Chaetoceros didymus</i>	14
	<i>Chaetoceros eibonii</i>	2
	<i>Chaetoceros lauderi</i>	5
	<i>Chaetoceros messanensis</i>	6
	<i>Chaetoceros socialis</i>	3
	Family.....Rhizosoleniaceae	
	<i>Rhizosolenia alata</i>	5
	<i>Rhizosolenia delicatula</i>	1
	<i>Rhizosolenia fragilissima</i>	3
	<i>Rhizosolenia imbricata</i>	1
	<i>Rhizosolenia setigera</i>	1
	<i>Rhizosolenia sp.</i>	1
	<i>Rhizosolenia stolterfothii</i>	9
	Family.....Coscinodiscaceae	
	<i>Coscinodiscus centralis</i>	17
	<i>Coscinodiscus granii</i>	5
	Suborder...Biddulphiineae	
	Family.....Biddulphiaceae	
	<i>Hemiaulus hauckii</i>	3
	<i>Hemiaulus sinensis</i>	1
Division...Chlorophyta		
Class.....Chlorophyceae (green algae)		
Order.....Chlorococcales		
Family.....Hydrodictyaceae		
<i>Pediastrum simplex</i>	1	
Family.....Oocystaceae		
<i>Kirchneriella subsolitaria</i>	1	
Family.....Scenedesmaceae		
<i>Scenedesmus dimorphus</i>	2	
Order.....Volvocales		
<i>Unidentified green flagellates</i>	1	
Family.....Pyramimonadaceae		
<i>Pyramimonas sp. 1</i>	4	
Order.....Zygnemales		
Family.....Desmediaceae		
<i>Staurostrum paradoxum</i>	1	
Family.....Zygnemaceae		
<i>Mougeotia sp. 1 (narrow trichomes)</i>	1	

Systematic review

Choctawhatchee Bay, day, surface, 64 μ samples

No. of occurrences

Division...	Cyanophyta	
Class.....	Cyanophyceae (blue-green algae or cyanobacteria)	
Order.....	Chroococcales	
Family.....	Chroococcaceae	
	<i>Merismopedia aeruginea</i>	1
	<i>Merismopedia elegans</i>	1
	<i>Merismopedia thermale</i>	1
	<i>Synechococcus sp.</i>	2
Order.....	Nostocales	
Family.....	Nostocaceae	
	<i>Anabaena cylindrica</i>	1
	<i>Anabaena inaequalis</i>	8
	<i>Anabaena sp. 1</i>	2
	<i>Anabaena sp. 2</i>	1
	<i>Anabaena sp.</i>	4
	<i>Anabaena variabilis</i>	2
Family.....	Oscillatoriaceae	
	<i>Hydrocoleum lyngbyaceum</i>	1
	<i>Johannesbaptistica sp.</i>	2
	<i>Lyngbya sordida</i>	2
	<i>Microcoleus lyngbyaceus</i>	1
	<i>Oscillatoria sp. 3</i>	2
	<i>Oscillatoria tenuis</i>	1
Division...	Dinophyta	
Class.....	Dinophyceae (dinoflagellates)	
	Unidentified dinoflagellates	4
Order.....	Gymnodiniales	
Family.....	Gymnodiniaceae	
	<i>Amphidinium carteri</i>	3
	<i>Amphidinium sp.</i>	2
	<i>Gymnodinium coeruleum</i>	1
	<i>Gymnodinium sp. 1</i>	1
	<i>Gymnodinium sp.</i>	3
Family.....	Polykrikaceae	
	<i>Polykrikos sp.</i>	2
Order.....	Peridinales	
Family.....	Peridiniaceae	
	<i>Oxyhris marina</i>	1
	<i>Peridinium brochii</i>	1
	<i>Peridinium claudicans</i>	4
	<i>Peridinium conicum</i>	1
	<i>Peridinium crassipes</i>	12
	<i>Peridinium depressum</i>	2
	<i>Peridinium divergens</i>	3
	<i>Peridinium elegans</i>	1
	<i>Peridinium gracile</i>	2
	<i>Peridinium leonis</i>	3
	<i>Peridinium oblongum</i>	16
	<i>Peridinium sp.</i>	6
	<i>Peridinium venustum</i>	1
Family.....	Protoperidiniaceae	

<i>Diplopsalis lenticula</i>	1
<i>Diplopsalis</i> sp.	8
<i>Encysted dinoflagellate</i>	55
Order.....Dinophysiales	
Family.....Amphisoleniaceae	
<i>Amphisolenia</i> sp.	1
Family.....Dinophysiaceae	
<i>Dinophysis caudata</i> f. <i>acutiformis</i>	12
<i>Dinophysis caudata</i>	18
<i>Dinophysis ovum</i>	1
<i>Dinophysis caudata</i> v. <i>pedunculata</i>	15
<i>Ornithoceros</i> sp.	3
Order.....Gonyaulacales	
Family.....Ceratiaceae	
<i>Ceratium tripos</i> v. <i>atlanticum</i>	11
<i>Ceratium carriense</i>	1
<i>Ceratium contortum</i>	3
<i>Ceratium declinatum</i>	1
<i>Ceratium furca</i>	21
<i>Ceratium fusus</i>	54
<i>Ceratium macroceros</i> v. <i>gallicum</i>	1
<i>Ceratium hircus</i>	86
<i>Ceratium inflatum</i>	1
<i>Ceratium lineatum</i>	1
<i>Ceratium declinatum</i> f. <i>normale</i>	2
<i>Ceratium tripos</i> v. <i>ponticum</i>	12
<i>Ceratium</i> sp.	1
<i>Ceratium vultus</i> v. <i>sumatranum</i>	1
<i>Ceratium teres</i>	3
<i>Ceratium trichoceros</i>	60
<i>Ceratium tripos</i>	76
<i>Characium-like</i> sp.	1
Family.....Gonyaulacaceae	
<i>Gonyaulax</i> sp. 1	1
<i>Gonyaulax</i> sp.	2
<i>Gonyaulax diegensis</i>	2
<i>Phalocrama cuneus</i>	1
<i>Phalacroma</i> sp.	2
Family.....Pyrocystaceae	
<i>Pyrocystis pseudonoctiluca</i> f. <i>biconica</i>	1
<i>Pyrocystis</i> sp. 1	69
Family.....Pyrophacaceae	
<i>Pyrophacus</i> sp.	1
<i>Pyrophacus horologium</i>	2
Order.....Prorocentrales	
Family.....Prorocentraceae	
<i>Prorocentrum compressum</i>	4
<i>Prorocentrum gracile</i>	7
<i>Prorocentrum micans</i>	18
<i>Prorocentrum pyriformis</i>	1
<i>Prorocentrum</i> sp. 1	2
<i>Prorocentrum</i> sp.	1
<i>Prorocentrum triestinum</i>	1
Division...Chrysophyta	
Class.....Chrysophyceae (golden-brown algae)	
<i>Dictyocha fibula</i>	29
<i>Dictyocha fibula</i> f. <i>rhombica</i>	1
<i>Dictyocha</i> sp.	1
Order.....Chryomonadales	

Family.....	Ochromonadaceae	
	<i>Dinobryon sertularia</i>	8
	<i>Dinobryon sp.</i>	3
Class.....	Haptophyceae	
	<i>Haptophyceae (haptophyte flagellate)</i>	1
Division...	Xanthophyta	
Class.....	Xanthophyceae (yellow-green algae)	
	<i>Xanthophyceae (yellow green flagellate)</i>	2
Order.....	Tribonemales	
Family.....	Tribonemataceae	
	<i>Tribonema affine</i>	2
	<i>Tribonema sp.</i>	1
Division...	Bacillariophyta (diatoms)	
Class.....	Bacillariophyceae	
Order.....	Pennales	
Suborder...	Araphidineae	
Family.....	Diatomaceae	
	<i>Asterionella formosa</i>	2
	<i>Asterionella japonica</i>	5
	<i>Ctenophora pulchella</i>	1
	<i>Delphineis livingstonii</i>	2
	<i>Delphineis surirella</i>	17
	<i>Dimerogramma marina</i>	7
	<i>Dimerogramma minor</i>	12
	<i>Falcula hyalina</i>	39
	<i>Falcula media</i>	1
	<i>Fragilaria capucina</i>	3
	<i>Fragilaria construens</i>	1
	<i>Fragilaria crotonensis</i>	1
	<i>Fragilaria sp.</i>	3
	<i>Fragilaria pinnata</i>	2
	<i>Grammatophora angulosa</i>	1
	<i>Grammatophora oceanica v. macilenta</i>	4
	<i>Grammatophora marina</i>	41
	<i>Grammatophora oceanica</i>	3
	<i>Grammatophora sp.</i>	1
	<i>Licmophora abbreviata</i>	5
	<i>Licmophora sp.</i>	1
	<i>Neodelphineis pelagica</i>	2
	<i>Opephora martyi</i>	20
	<i>Opephora pacifica</i>	6
	<i>Opephora schwartzii</i>	30
	<i>Opephora sp.</i>	3
	<i>Plagiogramma pulchellum</i>	5
	<i>Plagiogramma pulchellum v. pygmaea</i>	1
	<i>Plagiogramma sp.</i>	1
	<i>Podcystis adriaticum</i>	1
	<i>Rhabdonema adriaticum</i>	93
	<i>Rhaphoneis amphiceros</i>	1
	<i>Rhaphoneis liburnica</i>	2
	<i>Rhaphoneis sp.</i>	1
	<i>Striatella interrupta</i>	3
	<i>Striatella unipunctata</i>	46
	<i>Synedra acus</i>	6
	<i>Synedra affinis</i>	1
	<i>Synedra ulna v. amphirhyncus</i>	3
	<i>Synedra crystallina</i>	2
	<i>Synedra formosa</i>	1
	<i>Synedra gaillonii</i>	2

<i>Synedra tabulata</i> v. <i>grandis</i>	1
<i>Synedra hennedyana</i>	1
<i>Synedra ulna</i> v. <i>oxyrhy. f. mediacontracta</i>	1
<i>Synedra ulna</i> v. <i>oxyrhynchus</i>	11
<i>Synedra acus</i> v. <i>radians</i>	1
<i>Synedra tabulata</i>	24
<i>Synedra toxoneides</i>	1
<i>Synedra ulna</i>	18
<i>Synedra undulata</i>	2
<i>Tabularia investiens</i>	1
<i>Thalassionema nitzschioides</i>	43
<i>Thalassionema</i> sp.	2
<i>Thalassiothrix frauenfeldii</i>	24
<i>Thalassiothrix longissima</i>	2
<i>Thalassiothrix mediterranea</i>	3
<i>Thalassiothrix mediterranea</i> v. <i>pacifica</i>	1
Family.....Protoraphidaceae	
<i>Pseudohymantidium</i> sp.	1
Suborder...Raphidineae	
Family.....Eunotiaceae	
<i>Eunotia alpina</i>	4
<i>Eunotia bidentula</i>	1
<i>Eunotia exigua</i>	1
<i>Eunotia lunaris</i>	1
<i>Eunotia monodon</i>	2
<i>Eunotia pectinalis</i>	11
<i>Eunotia praerupta</i>	1
<i>Eunotia</i> sp.	4
Family.....Achnanthaceae	
<i>Achnanthes brevipes</i>	8
<i>Achnanthes clevei</i>	1
<i>Achnanthes lanceolata</i> v. <i>dubia</i>	1
<i>Achnanthes exigua</i>	2
<i>Achnanthes haukiana</i>	2
<i>Achnanthes</i> sp.	6
<i>Anorthoneis excentrica</i>	1
<i>Cocconeis placentula</i> v. <i>euglypta</i>	1
<i>Cocconeis placentula</i>	1
<i>Cocconeis scutellum</i>	60
<i>Cocconeis</i> sp.	1
Family.....Naviculaceae	
<i>Amphipleura pellucida</i>	1
<i>Amphiprora alata</i>	3
<i>Amphiprora gigantea</i>	26
<i>Amphiprora alata</i> f. <i>minor</i>	1
<i>Amphiprora pulchra</i>	3
<i>Amphiprora sulcata</i>	2
<i>Amphiprora</i> sp. 1	1
<i>Amphiprora</i> sp.	1
<i>Amphiprora gigantea</i> v. <i>sulcata</i>	1
<i>Amphora alata</i>	1
<i>Amphora angulosa</i>	1
<i>Amphora arcus</i>	2
<i>Amphora arenaria</i>	1
<i>Amphora cingulata</i>	2
<i>Amphora coffeaeformis</i>	3
<i>Amphora commutata</i>	3
<i>Amphora ocellata</i> v. <i>elongata</i>	1
<i>Amphora gigantea</i> v. <i>fusca</i>	1
<i>Amphora gigantea</i>	1

<i>Amphora holsatica</i>	1
<i>Amphora proteus</i> v. <i>maxima</i>	1
<i>Amphora mexicana</i>	1
<i>Amphora obtusa</i> v. <i>oceanica</i>	1
<i>Amphora obtusa</i>	15
<i>Amphora ocellata</i>	3
<i>Amphora ostrearia</i>	1
<i>Amphora ovalis</i>	8
<i>Amphora pediculus</i>	2
<i>Amphora proteus</i>	9
<i>Amphora obtusa</i> v. <i>rectangulata</i>	1
<i>Amphora</i> sp. 1	2
<i>Amphora</i> sp. 2	3
<i>Amphora</i> sp. 3	1
<i>Amphora</i> sp. 4	1
<i>Amphora</i> sp.	9
<i>Amphora</i> sp. (small)	1
<i>Amphora valida</i>	1
<i>Caloneis</i> sp. r265	1
<i>Caloneis acutiascula</i>	1
<i>Caloneis branderi</i>	1
<i>Caloneis latiascula</i>	2
<i>Caloneis libes</i>	2
<i>Caloneis maxima</i>	1
<i>Caloneis</i> sp.	2
<i>Caloneis westii</i>	1
<i>Capartogramma crucicula</i>	2
<i>Cymbella affinis</i>	1
<i>Cymbella aspera</i>	1
<i>Cymbella</i> sp.	3
<i>Diploneis adrena</i>	1
<i>Diploneis bombos</i>	2
<i>Diploneis crabro</i>	4
<i>Diploneis</i> sp.	1
<i>Diploneis ovalis</i> v. <i>oblongella</i>	1
<i>Diploneis ovalis</i>	16
<i>Diploneis puella</i>	1
<i>Diploneis smithii</i>	17
<i>Frustulia rhomboides</i>	1
<i>Gomphonema angustatum</i>	4
<i>Gomphonema constrictum</i>	1
<i>Gomphonema gracile</i>	1
<i>Gyrosigma acuminatum</i>	3
<i>Gyrosigma attenuatum</i>	1
<i>Gyrosigma balticum</i>	1
<i>Gyrosigma simile</i>	1
<i>Gyrosigma</i> sp.	6
<i>Mastogloia angulata</i>	14
<i>Mastogloia apiculata</i>	4
<i>Mastoneis biformis</i>	3
<i>Mastogloia braunii</i>	2
<i>Mastogloia cyclops</i>	2
<i>Mastogloia erythraea</i>	3
<i>Mastogloia hustedtii</i>	3
<i>Mastogloia lanceolata</i>	1
<i>Mastogloia macdonaldii</i>	1
<i>Mastogloia omissa</i>	1
<i>Mastogloia ovalis</i>	1
<i>Mastogloia portierana</i>	1
<i>Mastogloia rhombica</i>	1

<i>Mastogloia</i> sp.	3
<i>Mastogloia suboculata</i>	1
<i>Mastogloia varians</i>	1
<i>Navicula ambigua</i> -like	1
<i>Navicula</i> cf. <i>ammophila</i>	3
<i>Navicula bailyana</i>	3
<i>Navicula bioculata</i>	1
<i>Navicula clavata</i>	6
<i>Navicula granulata</i> v. <i>constricta</i>	1
<i>Navicula cryptocephala</i>	3
<i>Navicula directa</i>	7
<i>Navicula distans</i>	1
<i>Navicula irroratoides</i> f. <i>elliptica</i>	1
<i>Navicula forcipata</i>	1
<i>Navicula gracilis</i>	1
<i>Navicula gregaria</i>	2
<i>Navicula granulata</i>	6
<i>Navicula henneydi</i>	1
<i>Navicula irrorata</i>	2
<i>Navicula lyra</i>	17
<i>Navicula marina</i>	1
<i>Navicula monilifera</i>	1
<i>Navicula pinnata</i>	1
<i>Navicula pupula</i>	1
<i>Navicula scopularm</i>	2
<i>Navicula</i> sp. 1	3
<i>Navicula</i> sp. 2	3
<i>Navicula</i> sp. 3	2
<i>Navicula</i> sp. 4	2
<i>Navicula</i> sp.	9
<i>Navicula</i> sp. (very small)	1
<i>Navicula viridula</i>	2
<i>Navicula yarrensensis</i> v. <i>americana</i>	8
<i>Navicula yarrensensis</i>	33
<i>Navicula zanardiniana</i>	1
<i>Pinnularia gibba</i>	1
<i>Pinnularia</i> sp.	1
<i>Pinnularia viridis</i>	1
<i>Pleurosigma balticum</i>	1
<i>Pleurosigma delicatum</i>	1
<i>Pleurosigma elongatum</i>	4
<i>Pleurosigma formosum</i>	11
<i>Pleurosigma naviculaceum</i>	1
<i>Pleurosigma</i> sp. 2	1
<i>Pleurosigma speciosum</i>	1
<i>Pleurosigma</i> sp.	4
<i>Pleurosigma</i> sp. 1	1
<i>Pleurosigma sulcata</i>	1
<i>Stauroneis anceps</i>	1
<i>Stauroneis pachycephala</i>	3
<i>Stauroneis smithii</i>	1
<i>Stauroneis spicula</i>	6
<i>Trachyneis aspera</i>	1
<i>Tropidoneis lepidoptera</i>	8
<i>Tropidoneis</i> sp.	1
Family.....Epithemiaceae	
<i>Rhopalodia gibba</i>	4
<i>Rhopalodia gibberula</i>	4
Family.....Nitzschiaceae	
<i>Bacillaria paradoxa</i>	23

<i>Bacillaria paxillifer</i>	20
<i>Hantzschia</i> sp.	2
<i>Nitzschia acuminata</i>	1
<i>Nitzschia angularis</i>	1
<i>Nitzschia circumsuta</i>	1
<i>Nitzschia acicularis</i> v. <i>closterioides</i>	8
<i>Nitzschia clarissima</i>	1
<i>Nitzschia coarctata</i>	1
<i>Nitzschia constricta</i>	2
<i>Nitzschia closterium</i>	14
<i>Nitzschia delicatissima</i>	1
<i>Nitzschia gartlopii</i>	1
<i>Nitzschia sigma</i> v. <i>habirshawii</i>	1
<i>Nitzschia incurva</i>	1
<i>Nitzschia insignis</i>	3
<i>Nitzschia jelinecki</i>	1
<i>Nitzschia lanceolata</i>	3
<i>Nitzschia longa</i>	10
<i>Nitzschia longissima</i>	17
<i>Nitzschia lorenziana</i>	7
<i>Nitzschia macilenta</i>	1
<i>Nitzschia maxima</i>	2
<i>Nitzschia obtusa</i>	5
<i>Nitzschia palea</i>	2
<i>Nitzschia panduriformis</i>	4
<i>Nitzschia paradoxa</i>	1
<i>Nitzschia punctata</i>	3
<i>Nitzschia pungens</i>	3
<i>Nitzschia longissima</i> v. <i>reversa</i>	2
<i>Nitzschia rigida</i>	1
<i>Nitzschia lorenziana</i> v. <i>subtilis</i>	1
<i>Nitzschia scalaris</i>	2
<i>Nitzschia seriata</i>	7
<i>Nitzschia sigma</i>	26
<i>Nitzschia sigma</i> v. <i>sigmatella</i>	1
<i>Nitzschia sigmoidea</i>	5
<i>Nitzschia socialis</i>	1
<i>Nitzschia</i> sp. 1	1
<i>Nitzschia</i> sp. 2	1
<i>Nitzschia</i> sp. (arc shaped)	1
<i>Nitzschia</i> sp.	15
<i>Nitzschia tryblionella</i> v. <i>subsalina</i>	1
<i>Nitzschia triblionella</i> v. <i>salinarum</i>	1
<i>Nitzschia triblionella</i>	10
<i>Nitzschia longissima</i> v. <i>typica</i>	1
<i>Nitzschia valida</i>	2
<i>Pseudoeunotia doliolus</i>	1
<i>Pseudoeunotia</i> sp.	1
Family..... <i>Surirellaceae</i>	
<i>Campylodiscus clypeus</i>	5
<i>Campylodiscus echeineis</i>	4
<i>Campylodiscus eximius</i>	2
<i>Campylodiscus imperialis</i>	2
<i>Campylodiscus limbatus</i>	7
<i>Campylodiscus samoensis</i>	1
<i>Campylodiscus</i> sp.	1
<i>Surirella</i> sp. r265	1
<i>Surirella fastuosa</i> v. <i>cuneata</i>	1
<i>Surirella fastuosa</i>	11
<i>Surirella gemma</i>	6

<i>Surirella robusta f. minor</i>	1
<i>Surirella tenera v. nervosa</i>	1
<i>Surirella ovata</i>	3
<i>Surirella ovalis</i>	2
<i>Surirella fastuosa v. recedens</i>	1
<i>Surirella robusta</i>	3
<i>Surirella sp. 1</i>	1
<i>Surirella sp.</i>	1
<i>Surirella tenera</i>	3
Order.....Centrales	
Suborder...Coscinodiscineae	
Family.....Thalassiosiraceae	
<i>Aulacosira granulata v. angustissima</i>	1
<i>Aulacosira granulata</i>	8
<i>Cyclotella meneghiniana</i>	38
<i>Cyclotella sp. 1</i>	3
<i>Cyclotella sp. 2</i>	1
<i>Cyclotella sp.</i>	7
<i>Cyclotella sp. (small)</i>	97
<i>Cyclotella striata</i>	88
<i>Cyclotella stylorum</i>	8
<i>Lauderia borealis</i>	1
<i>Lauderia compressa</i>	1
<i>Skeletonema costatum</i>	19
<i>Thalassiosira decipiens</i>	9
<i>Thalassiosira eccentrica</i>	26
<i>Thalassiosira gracilis</i>	3
<i>Thalassiosira lineatus</i>	3
<i>Thalassiosira nannolineata</i>	4
<i>Thalassiosira oestrupii</i>	40
<i>Thalassiosira sp. 1</i>	1
<i>Thalassiosira sp. 2</i>	1
<i>Thalassiosira sp.</i>	13
<i>Thalassiosira sp. (small)</i>	4
Family.....Melosiraceae	
<i>Corethron hystrix</i>	4
<i>Corethron pelagicum</i>	1
<i>Hyalodiscus radiatus</i>	3
<i>Leptocylindrus danicus</i>	26
<i>Leptocylindrus-like</i>	1
<i>Melosira (aulacosira) granulata</i>	1
<i>Melosira ambigua</i>	1
<i>Melosira borresi</i>	1
<i>Melosira dubia</i>	6
<i>Melosira granulata</i>	4
<i>Melosira juergensii</i>	1
<i>Melosira moniliformis</i>	2
<i>Melosira undulata v. moranii</i>	1
<i>Melosira undulata v. normanii</i>	1
<i>Melosira nummuloides</i>	4
<i>Melosira sp.</i>	3
<i>Melosira undulata</i>	2
<i>Melosira varians</i>	16
<i>Paralia sulcata v. biseriata</i>	2
<i>Paralia sulcata f. coronata</i>	4
<i>Paralia sulcata</i>	46
<i>Podosira stelliger</i>	3
<i>Stephanopyxis turris</i>	5
Family.....Hemidiscaceae	
<i>Actinocyclus crassus</i>	1

<i>Actinocyclus curvatulus</i>	1
<i>Actinocyclus ehrenbergii</i>	58
<i>Actinocyclus ehrenbergii v. ralfsii</i>	5
<i>Actinocyclus ralfsii</i>	2
<i>Actinocyclus ralfsii v. sparsus</i>	1
<i>Actinocyclus sp.</i>	4
<i>Actinocyclus tenuissimus</i>	1
<i>Actinocyclus ehrenbergii v. tenella</i>	2
<i>Hemidiscus cuneiformis</i>	1
Family.....Heliopeltaceae	
<i>Actinoptychus senarius</i>	2
<i>Actinoptychus undulatus</i>	30
Family.....Asterolampraceae	
<i>Asteromphalus flabellatus</i>	3
<i>Asteromphalus heptactis</i>	1
Family.....Eupodiscaceae	
<i>Auliscus caelatus</i>	1
<i>Triceratium favus</i>	2
<i>Triceratium sculptum</i>	1
<i>Triceratium spinosum</i>	1
<i>Triceratium sp.</i>	1
Family.....Chaetoceraceae	
<i>Bacteriastrum delicatulum</i>	2
<i>Bacteriastrum elongatum</i>	2
<i>Bacteriastrum hyalinum</i>	34
<i>Bacteriastrum hyalinum v. princeps</i>	1
<i>Bacteriastrum varians</i>	15
<i>Chaetoceros affinis</i>	15
<i>Chaetoceros atlanticus</i>	1
<i>Chaetoceros didymus v. atlanticus</i>	1
<i>Chaetoceros brevis</i>	75
<i>Chaetoceros convolutus</i>	1
<i>Chaetoceros coarctatus</i>	42
<i>Chaetoceros compressus</i>	7
<i>Chaetoceros constrictus</i>	5
<i>Chaetoceros costatum</i>	4
<i>Chaetoceros crinitus</i>	1
<i>Chaetoceros curvisetus</i>	8
<i>Chaetoceros danicus</i>	2
<i>Chaetoceros debile</i>	3
<i>Chaetoceros decipiens</i>	97
<i>Chaetoceros densus</i>	20
<i>Chaetoceros didymus</i>	90
<i>Chaetoceros diversus</i>	1
<i>Chaetoceros eibonii</i>	10
<i>Chaetoceros filiformis</i>	1
<i>Chaetoceros furcellatus</i>	3
<i>Chaetoceros gracilis</i>	10
<i>Chaetoceros lacinosus</i>	38
<i>Chaetoceros lauderi</i>	24
<i>Chaetoceros lorenzianus</i>	31
<i>Chaetoceros mediterranea</i>	1
<i>Chaetoceros messanensis</i>	16
<i>Chaetoceros pelagicus</i>	10
<i>Chaetoceros pendulus</i>	2
<i>Chaetoceros didymus v. protuberans</i>	32
<i>Chaetoceros pseudocurvisetus</i>	6
<i>Chaetoceros radicans</i>	1
<i>Chaetoceros socialis</i>	77
<i>Chaetoceros sp.</i>	1

<i>Chaetoceros wigami</i>	1
<i>Cymatosira belgica</i>	2
<i>Cymatosira lorenziana</i>	19
Family.....Lithodesmiaceae	
<i>Ditylum brightwelli</i>	2
<i>Lithodesmium undulatum</i>	3
<i>Streptotheca indica</i>	1
Family.....Rhizosoleniaceae	
<i>Guinardia flaccida</i>	11
<i>Rhizosolenia alata</i>	23
<i>Rhizosolenia calcaravis</i>	9
<i>Rhizosolenia delicatula</i>	5
<i>Rhizosolenia fragilissima</i>	6
<i>Rhizosolenia alata f. gracillima</i>	2
<i>Rhizosolenia hebetata</i>	6
<i>Rhizosolenia imbricata</i>	13
<i>Rhizosolenia alata f. indica</i>	2
<i>Rhizosolenia alata v. inervis</i>	1
<i>Rhizosolenia styliformis v. longispina</i>	1
<i>Rhizosolenia robusta</i>	2
<i>Rhizosolenia hebetata v. semispina</i>	2
<i>Rhizosolenia setigera</i>	8
<i>Rhizosolenia imbricata v. shrubsolei</i>	2
<i>Rhizosolenia stolterfothii</i>	54
<i>Rhizosolenia styliformis</i>	8
Family.....Coscinodiscaceae	
<i>Coscinodiscus apiculatus v. ambigua</i>	1
<i>Coscinodiscus apiculatus</i>	4
<i>Coscinodiscus argus</i>	1
<i>Coscinodiscus asteromphalus</i>	4
<i>Coscinodiscus centralis</i>	154
<i>Coscinodiscus concinnus</i>	1
<i>Coscinodiscus curvatulus</i>	4
<i>Coscinodiscus divisus</i>	1
<i>Coscinodiscus granii</i>	123
<i>Coscinodiscus janischii</i>	1
<i>Coscinodiscus marginatus</i>	3
<i>Coscinodiscus obscurus</i>	3
<i>Coscinodiscus oculus-iridis</i>	8
<i>Coscinodiscus operculatus</i>	2
<i>Coscinodiscus perforatus v. pavillardii</i>	2
<i>Coscinodiscus perforatus</i>	12
<i>Coscinodiscus radiatus</i>	5
<i>Coscinodiscus sp.</i>	4
<i>Psammodiscus nitidus</i>	3
<i>Stellarima microtrias</i>	1
Suborder...Biddulphiineae	
Family.....Biddulphiaceae	
<i>Anaulus sp.</i>	1
<i>Attheya decora</i>	1
<i>Biddulphia laevis</i>	1
<i>Biddulphia mobiliensis</i>	5
<i>Biddulphia regia</i>	1
<i>Biddulphia sinensis</i>	5
<i>Ethmodiscus sp.</i>	1
<i>Eucampia zoodiacus</i>	2
<i>Eunatogramma laevis</i>	1
<i>Eunatogramma weissei</i>	2
<i>Hemiaulus hauckii</i>	19
<i>Hemiaulus membranacea</i>	1

	<i>Hemiaulus sinensis</i>	3
	<i>Hydrosera whampoensis</i>	1
	<i>Terpsinoe americana</i>	1
	<i>Terpsinoe intermedia</i>	1
Division...	Chlorophyta	
Class.....	Chlorophyceae (green algae)	
Order.....	Chlorococcales	
	Family.....Hydrodictyaceae	
	<i>Pediastrum biradiatum</i>	3
	<i>Pediastrum simplex</i> v. <i>duodenarium</i>	2
	<i>Pediastrum simplex</i>	5
	Family.....Scenedesmaceae	
	<i>Scenedesmus quadricauda</i>	1
Order.....	Ulotrichales	
	Family.....Microsporaceae	
	<i>Microspora</i> sp. 1	1
	<i>Microspora</i> sp. 2	1
	<i>Microspora</i> sp.	1
Order.....	Volvocales	
	<i>Unidentified green flagellates</i>	1
	Family.....Volvocaceae	
	<i>Volvox aureus</i>	1
Order.....	Zygnemales	
	Family.....Desmediaceae	
	<i>Closterium</i> sp. 1	1
	<i>Closterium</i> sp. 2	1
	<i>Microsterias</i> sp.	1
	<i>Staurastrum notatum</i>	1
	<i>Staurastrum paradoxum</i>	2
	Family.....Zygnemaceae	
	<i>Mougeotia</i> sp. 1 (narrow trichomes)	2
	<i>Mougeotia</i> sp. 2 (wide trichomes)	1
	<i>Mougeotia</i> sp. 3	1
	<i>Mougeotia</i> sp. 4	2
	<i>Mougeotia</i> sp.	3
Systematics unknown or unassigned		
	<i>Dissodinium</i> sp.	1
	<i>Unidentified flagellates</i>	20

Systematic review

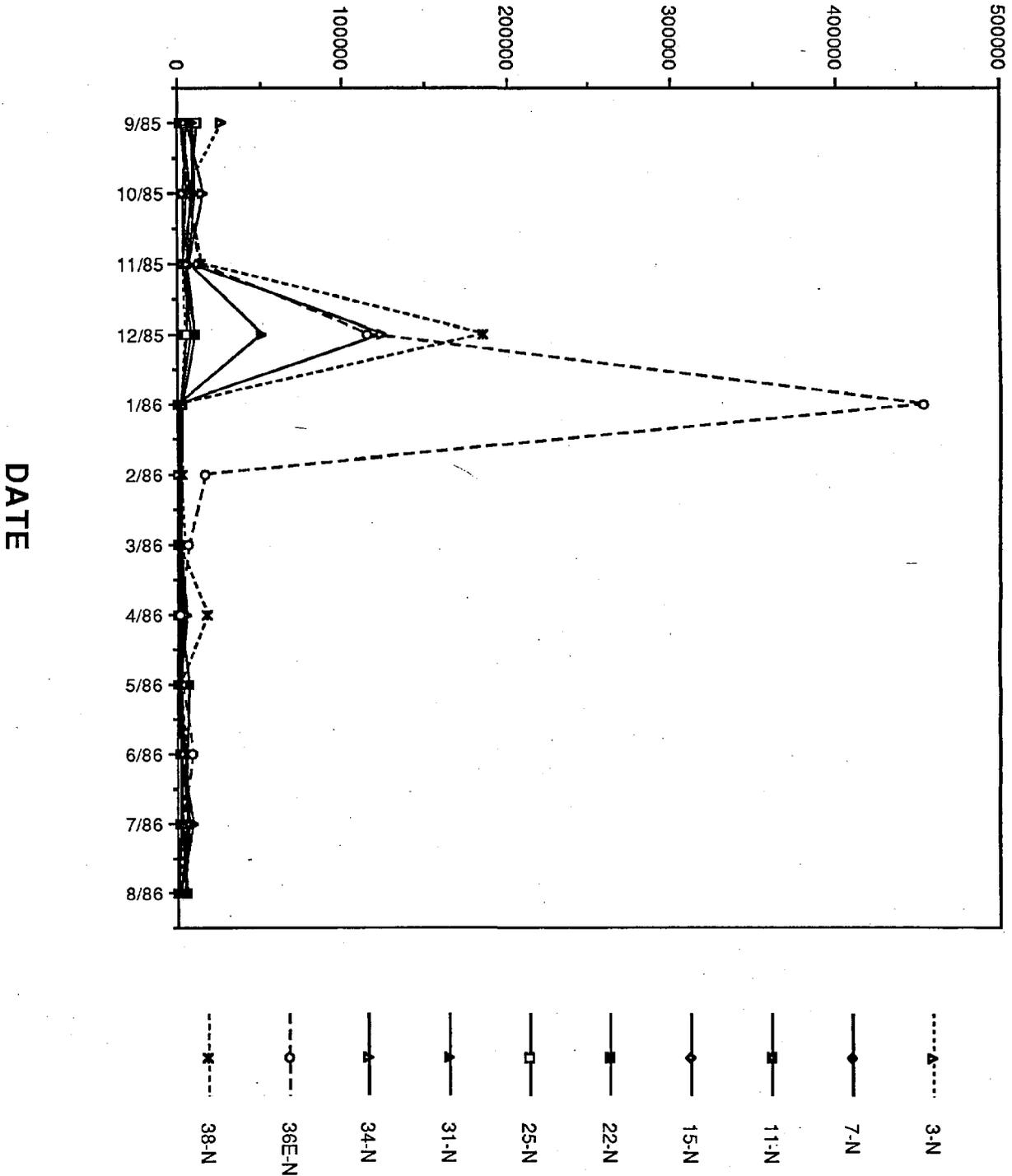
Offshore (gulf) stations, day, surface

	No. of occurrences
Division...Cyanophyta	
Class.....Cyanophyceae (blue-green algae or cyanobacteria)	
Order.....Chroococcales	
Family.....Chroococcaceae	
<i>Eucapsis</i> sp.	2
Order.....Nostocales	
Family.....Oscillatoriaceae	
<i>Microcoleus lyngbyaceus</i>	7
<i>Oscillatoria membranacea</i>	1
<i>Schizothrix</i> sp.	1
<i>Trichodesmium erythraeum</i>	7
Division...Dinophyta	
Class.....Dinophyceae (dinoflagellates)	
Order.....Peridinales	
Family.....Peridiniaceae	
<i>Peridinium conicum</i>	6
<i>Peridinium elegans</i>	1
<i>Peridinium nipponicum</i>	1
<i>Peridinium oblongum</i>	13
<i>Peridinium</i> sp.	2
Family.....Proto-peridiniaceae	
<i>Diplopsalis lenticula</i>	6
Order.....Dinophysiales	
Family.....Dinophysiaceae	
<i>Cladopyxis brachiolata</i>	2
<i>Dinophysis caudata</i>	7
<i>Ornithoceros magnificus</i>	2
<i>Oxytoxum</i> sp.	2
Order.....Gonyaulacales	
Family.....Ceratiaceae	
<i>Ceratium furca</i>	13
<i>Ceratium fusus</i>	9
<i>Ceratium hircus</i>	5
<i>Ceratium teres</i>	3
<i>Ceratium trichoceros</i>	12
<i>Ceratium tripos</i>	5
<i>Ceratium vultar</i>	1
Family.....Gonyaulacaceae	
<i>Gonyaulax monilata</i>	1
Family.....Pyrocystaceae	
<i>Noctiluca marina</i>	1
Family.....Pyrophacaceae	
<i>Pyrophacus horologium</i>	1
Family.....Ceratocoryaceae	
<i>Ceratocorys horrida</i>	4
Order.....Prorocentrales	
Family.....Prorocentraceae	
<i>Prorocentrum gracile</i>	1
<i>Prorocentrum micans</i>	2
<i>Prorocentrum minimum</i>	1
Division...Bacillariophyta (diatoms)	

Class.....	Bacillariophyceae	
Order.....	Pennales	
Suborder...	Araphidineae	
Family.....	Diatomaceae	
	<i>Asterionella</i> sp.	1
	<i>Asterionella japonica</i>	2
	<i>Delphineis livingstonii</i>	1
	<i>Delphineis surirella</i>	2
	<i>Dimerogramma marina</i>	1
	<i>Plagiogramma-like</i> sp.	2
	<i>Plagiogramma</i> sp.	1
	<i>Rhabdonema adriaticum</i>	5
	<i>Striatella unipunctata</i>	5
	<i>Thalassionema nitzschioides</i>	6
	<i>Thalassiothrix frauenfeldii</i>	13
	<i>Thalassiothrix longissima</i>	2
	<i>Thalassiothrix mediterranea</i>	2
	<i>Thalassiothrix mediterranea</i> v. <i>pacifica</i>	3
Suborder...	Raphidineae	
Family.....	Naviculaceae	
	<i>Amphiprora gigantea</i>	1
	<i>Amphora obtusa</i>	1
	<i>Gyrosigma macrum</i>	1
	<i>Haslea</i> sp.	1
	<i>Navicula granulata</i>	1
	<i>Navicula</i> sp. (needle type)	2
	<i>Pleurosigma angulatum</i>	5
Family.....	Nitzschiaceae	
	<i>Bacillaria paxillifer</i>	1
	<i>Nitzschia closterium</i>	3
	<i>Nitzschia longissima</i>	3
	<i>Nitzschia pungens</i>	3
	<i>Nitzschia seriata</i>	1
	<i>Nitzschia sigmoidea</i>	1
Family.....	Surirellaceae	
	<i>Campylodiscus echeineis</i>	1
Order.....	Centrales	
Suborder...	Coscinodiscineae	
Family.....	Thalassiosiraceae	
	<i>Cyclotella striata</i>	1
	<i>Skeletonema costatum</i>	11
	<i>Thalassiosira eccentrica</i>	6
	<i>Thalassiosira oestrupii</i>	4
Family.....	Melosiraceae	
	<i>Leptocylindrus danicus</i>	2
	<i>Paralia sulcata</i>	1
	<i>Podosira stelliger</i>	5
	<i>Stephanopyxis palmeriana</i>	6
	<i>Stephanopyxis turris</i>	3
Family.....	Hemidiscaceae	
	<i>Actinocyclus ehrenbergii</i>	3
	<i>Azpeitia nodulifer</i>	1
	<i>Hemidiscus cuneiformis</i>	3
	<i>Hemidiscus</i> sp.	4
Family.....	Heliopeltaceae	
	<i>Actinoptychus senarius</i>	2
Family.....	Eupodiscaceae	
	<i>Aulacodiscus argus</i>	3
	<i>Auliscus sculptus</i>	1

<i>Eupodiscus radiatus</i>	2
Family.....Chaetoceraceae	
<i>Bacteriastrum varians</i>	16
<i>Chaetoceros affinis</i>	3
<i>Chaetoceros atlanticus</i>	1
<i>Chaetoceros brevis</i>	10
<i>Chaetoceros coarctatus</i>	2
<i>Chaetoceros curvisetus</i>	2
<i>Chaetoceros decipiens</i>	8
<i>Chaetoceros densus</i>	1
<i>Chaetoceros didymus</i>	4
<i>Chaetoceros diversus</i>	8
<i>Chaetoceros eibonii</i>	1
<i>Chaetoceros lauderi</i>	1
<i>Chaetoceros lorenzianus</i>	1
<i>Chaetoceros messanensis</i>	1
<i>Chaetoceros peruvianus</i>	6
<i>Chaetoceros pseudocurvisetus</i>	1
<i>Chaetoceros teres</i>	1
<i>Cymatosira belgica</i>	5
<i>Cymatosira lorenziana</i>	2
Family.....Lithodesmiaceae	
<i>Bellorochea malleus</i>	1
<i>Ditylum brightwelli</i>	5
<i>Lithodesmium undulatum</i>	3
Family.....Rhizosoleniaceae	
<i>Corethron cryophyllum</i>	1
<i>Dactyliosolen</i> sp.	1
<i>Detonula</i> sp.	1
<i>Guinardia flaccida</i>	16
<i>Rhizosolenia alata</i>	15
<i>Rhizosolenia calcaravis</i>	6
<i>Rhizosolenia imbricata</i>	18
<i>Rhizosolenia alata</i> f. <i>indica</i>	1
<i>Rhizosolenia robusta</i>	15
<i>Rhizosolenia setigera</i>	11
<i>Rhizosolenia stolterfothii</i>	16
<i>Rhizosolenia styliformis</i>	6
<i>Rhizosolenia castracanii</i>	9
Family.....Coscinodiscaceae	
<i>Coscinodiscus centralis</i>	9
<i>Coscinodiscus concinnus</i>	3
<i>Coscinodiscus granii</i>	5
<i>Coscinodiscus jonesianus</i>	3
<i>Coscinodiscus operculatus</i>	1
<i>Coscinodiscus radiatus</i>	3
<i>Stellarima microtrias</i>	1
Suborder...Biddulphiineae	
Family.....Biddulphiaceae	
<i>Biddulphia mobiliensis</i>	4
<i>Biddulphia rhombus</i>	1
<i>Biddulphia sinensis</i>	10
<i>Biddulphia sinensis</i> -like	1
<i>Biddulphia titiana</i>	1
<i>Climacodium frauenfeldianum</i>	8
<i>Ditylum</i> sp.	1
<i>Eucampia cornuta</i>	1
<i>Eucampia zodiacus</i>	11
<i>Hemiaulus membranacea</i>	5
<i>Hemiaulus sinensis</i>	7

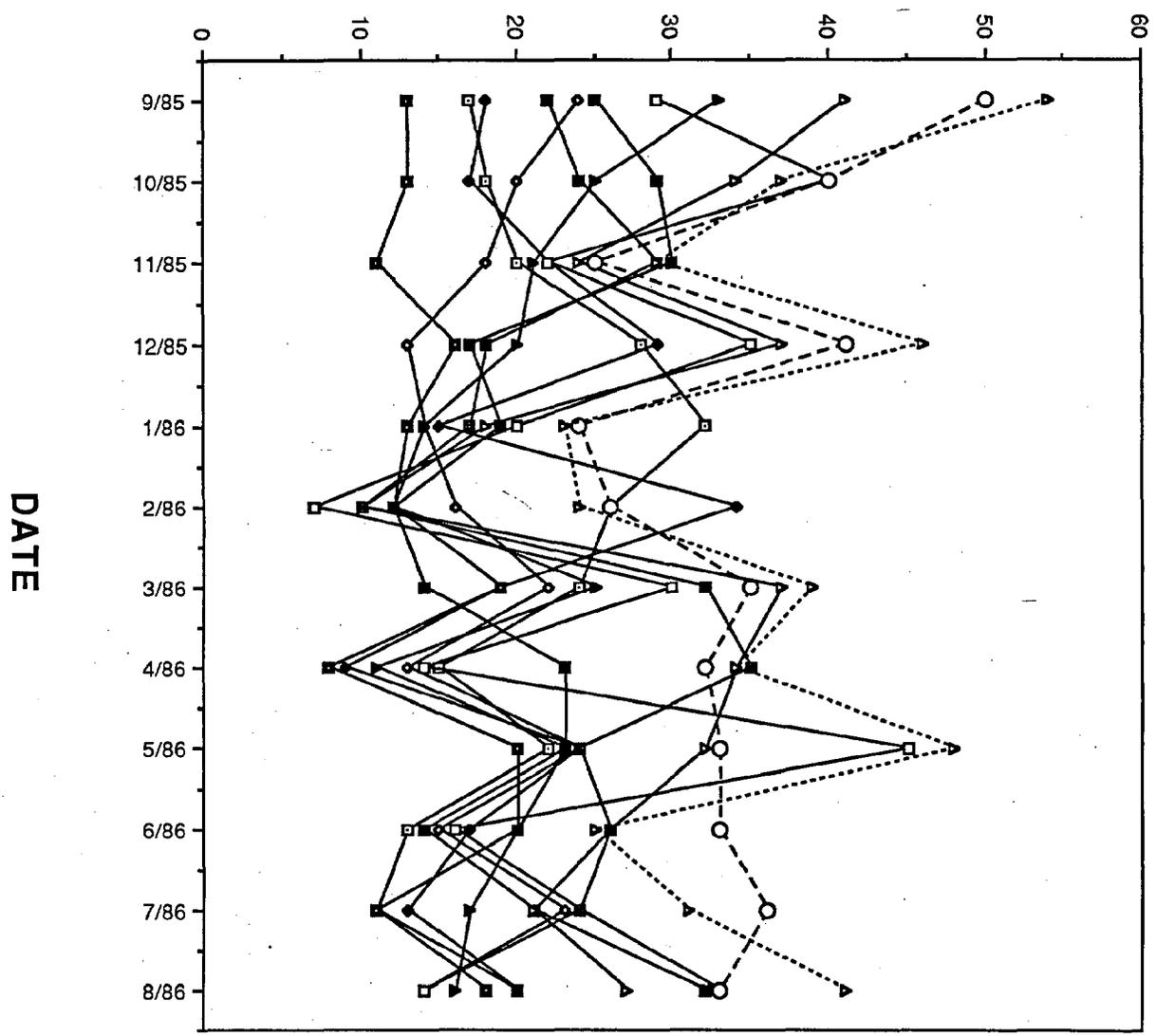
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diversity taken monthly from September, 1985 through August, 1986.

CHOCIAWHAICHEE BAY ALGAE: 64 MICRO-M

SPECIES RICHNESS

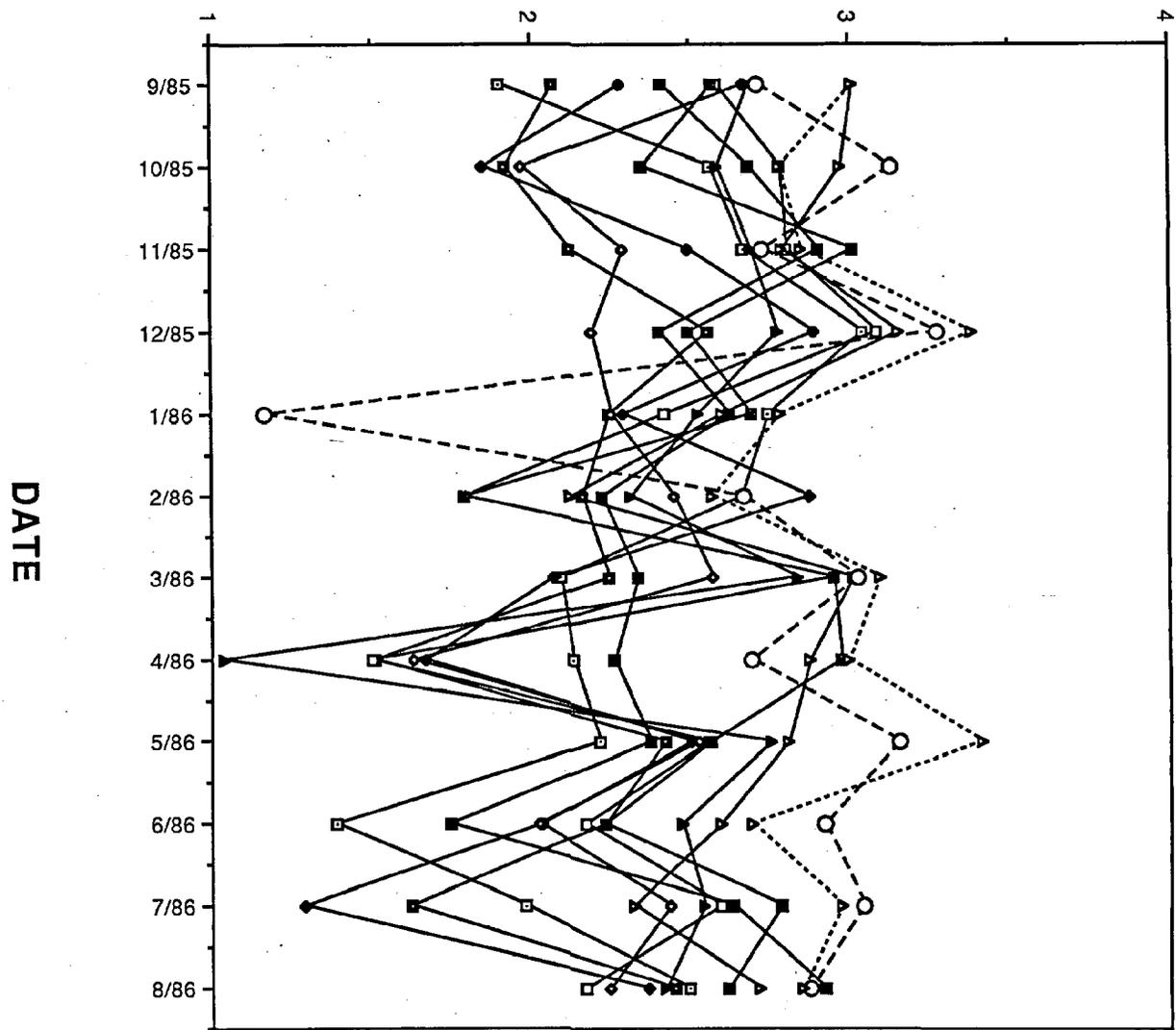


DATE

- 3-NSPP
- 7-NSPP
- 11-NSPP
- 15-NSPP
- 19-NSP
- 22-NSPP
- ▲— 25-NSPP
- ▲— 31-NSPP
- 34-NSP
- 36-NSPP
- ▲— 38-NSPP

CHOCATWATCHEE BAY ALGAE: 64 MICRO-M

SHANNON DIVERSITY



DATE

- 3-SHAN.DIV.
- 7-SHAN.DIV.
- 11-SHAN.DIV.
- 15-SHAN.DIV.
- 19-SHAN.DIV.
- 22-SHAN.DIV.
- ▲— 25-SHAN.DIV.
- ▲— 31-SHAN.DIV.
- 34-SHAN.DIV.
- 36E-SHAN.DIV.
- ▲— 38-SHAN.DIV.

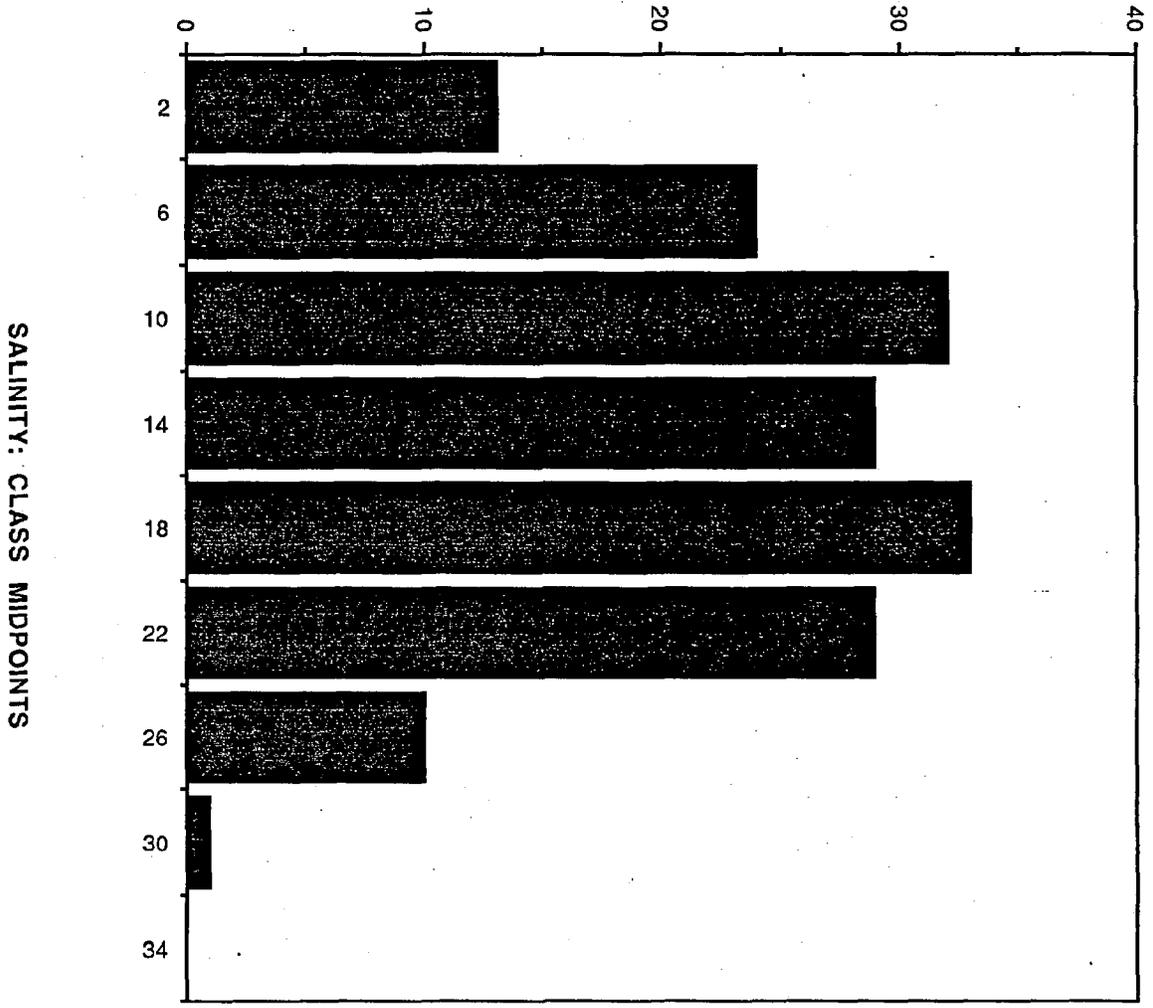
levels noted in winter months. Most of the other bay stations had relatively low species richness levels during most times of the year. With the exception of January, 1986 (the time of high numbers and high dominance), the species diversity was also highest at these two western stations. Such diversity indices were uniformly high at all other times of the year. In this way, eutrophicated conditions were characterized by high numbers of phytoplankton, high phytoplankton species richness and high diversity.

The dominant algal species in the Choctawhatchee Bay system over the period of study were run against the various important water quality factors as a distribution analysis by increments. The number of occurrences of specific ranges of important physical/chemical factors are shown graphically (Figure 8) along with the occurrences of dominant phytoplankton species along such gradients. Salinity levels in the bay were scattered across a relatively wide range with the highest frequencies of occurrence between 8 and 24 ppt. The phytoplankton species were located along gradients of salinity. For instance, *Chaetoceros brevis* was located along the mid-range of salinity whereas *C. didymus* was an indicator of higher salinities. Other species, such as *Skeletonema costatum*, were ranged along the lower levels of salinity. Most of the phytoplankton species showed specific ranges of salinity tolerance within varying ranges.

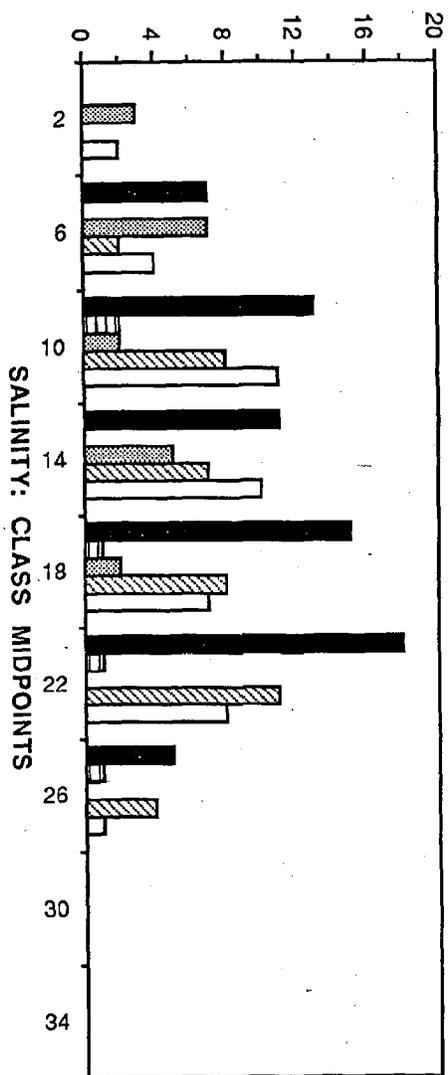
Dissolved oxygen (Figure 8) was relatively high at the surface of the bay throughout the year with levels remaining generally higher than 5 mg/l. Most of the phytoplankton were found at levels between 6.0 and 9.0 mg/l. The relative differences probably reflected seasonal changes in temperature and D. O. rather than any species-specific changes along D. O. gradients.

inure, ammonia, and chlorophyll a. The numerically dominant phytoplankton species (16) are tested as possible indicators of water quality phenomena in the Choctawhatchee Bay system from September, 1985 through August, 1986.

OCCURRENCES

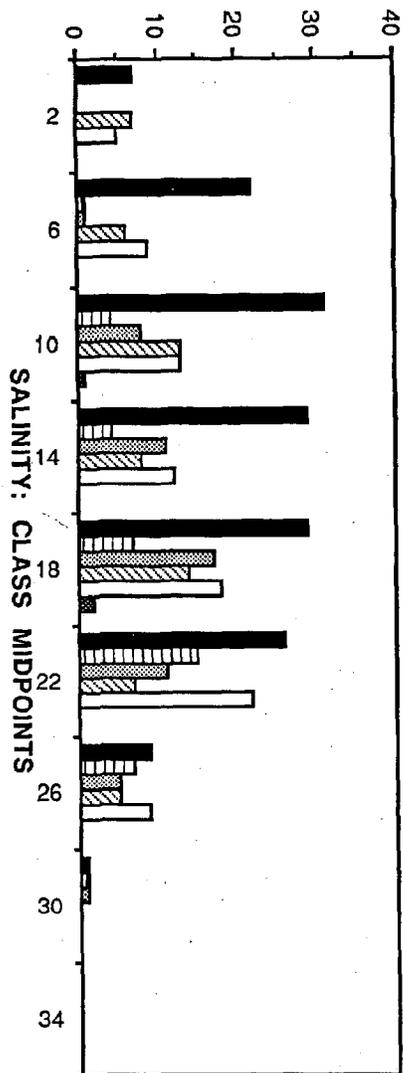


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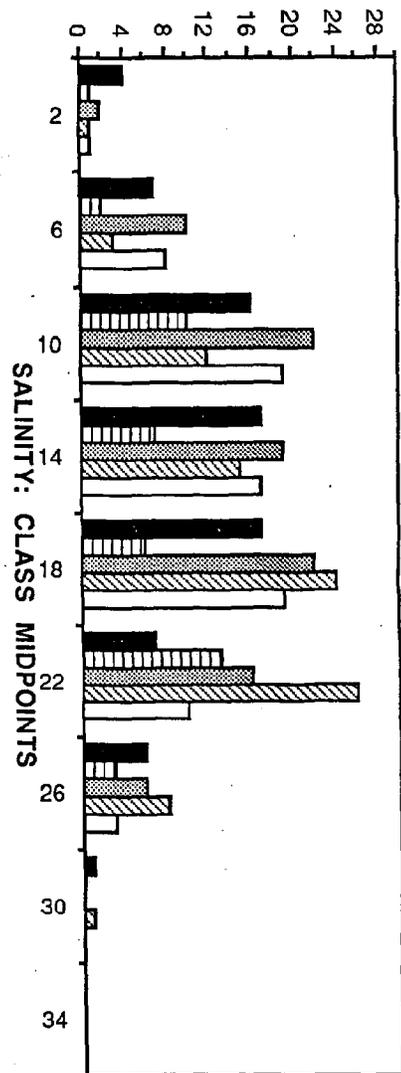
- Pyrocystis sp. 1
- ▨ Rhizosolenia delicatula
- ▩ Skeletonema costatum
- ▧ Thalassiosira oestakupii
- Thalassionema nitzschoides

OCCURRENCES



- Coscinodiscus centralis
- ▨ Falcula hyalina
- ▩ Rhizosolenia stolterfothii
- ▧ Cocconeis scutellum
- ▦ Cyclotella striata
- ▥ Navicula cf. armmophila

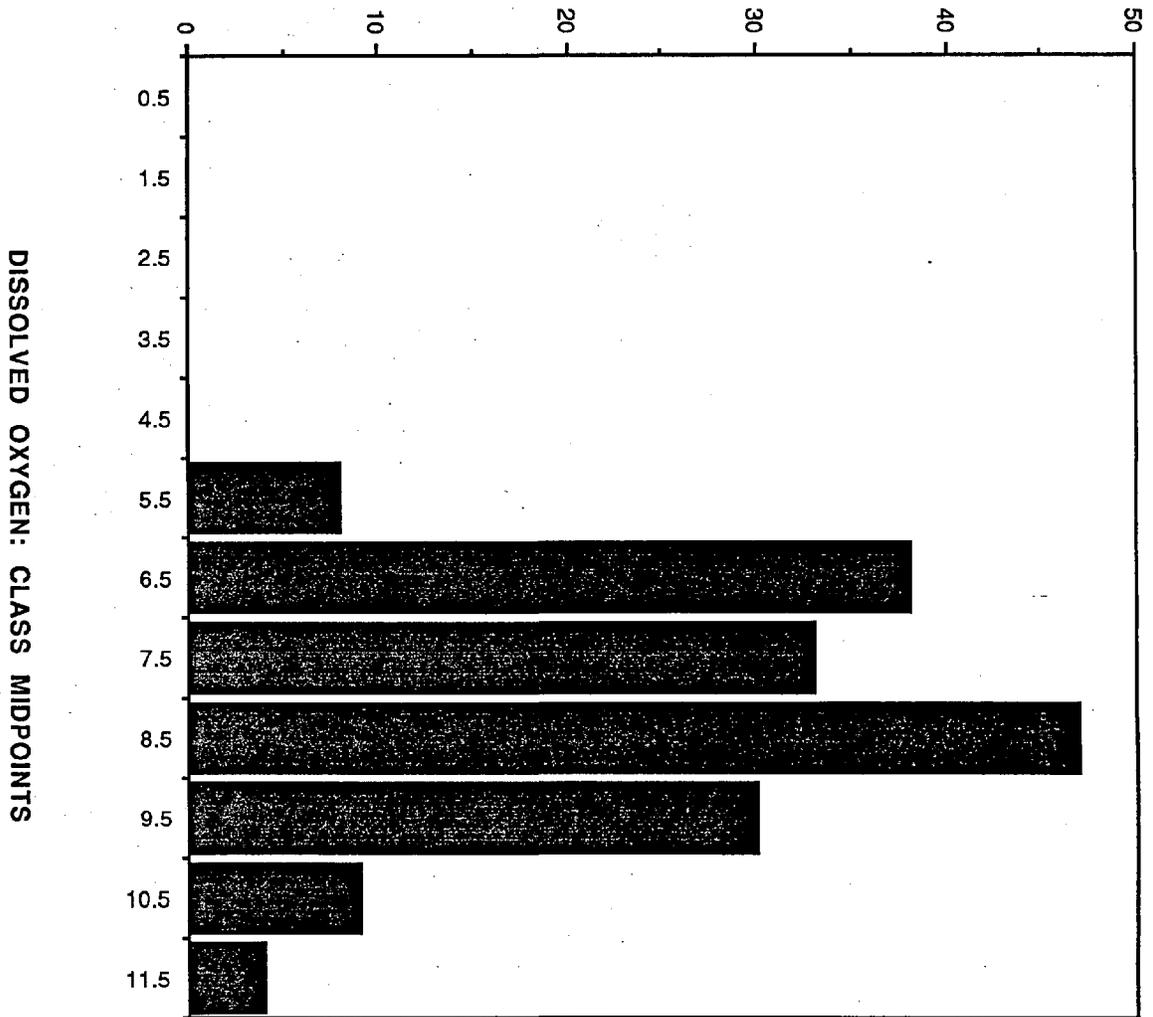
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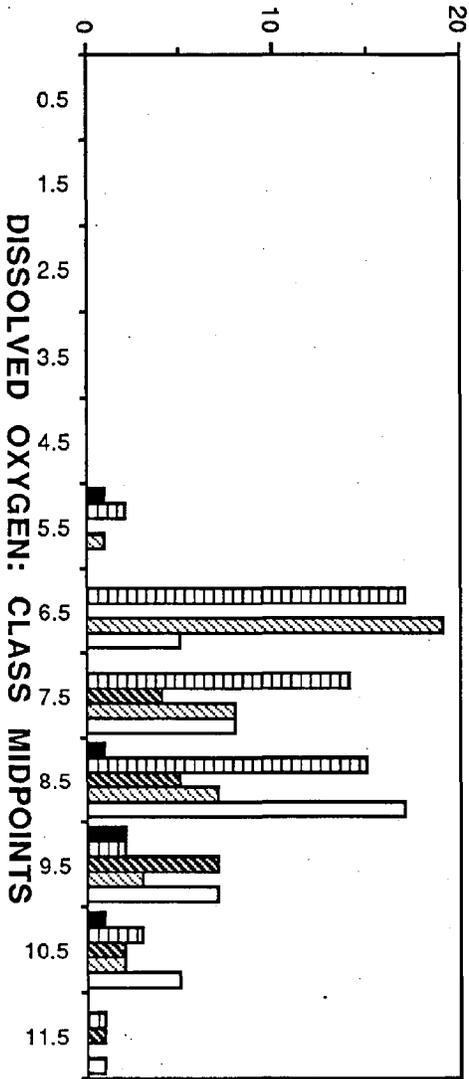
- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▩ Chaetoceros decipiens
- ▧ Chaetoceros didymus
- ▦ Chaetoceros socialis

CHOCTAWHATCHEE BAY 64 MICRO-M SAMPLES

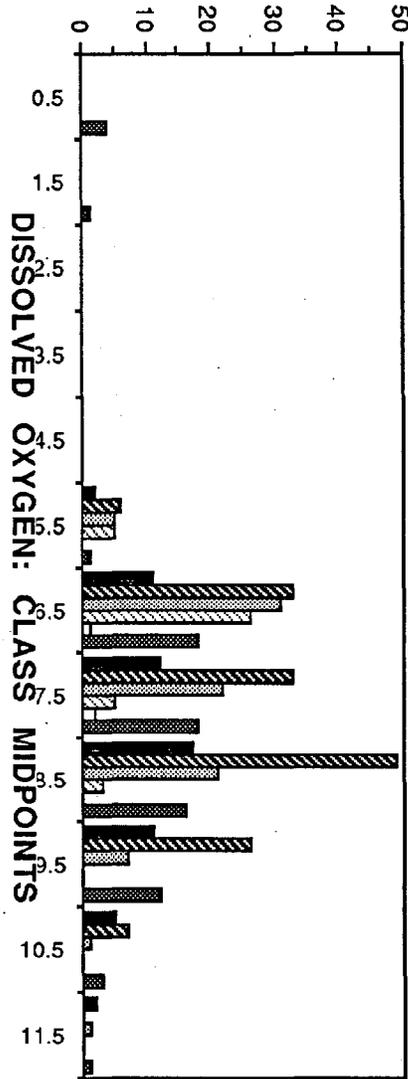
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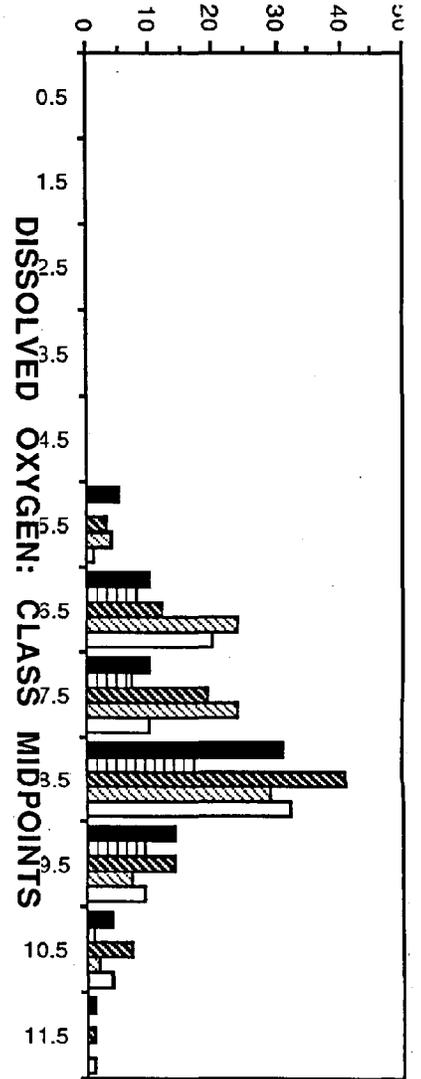
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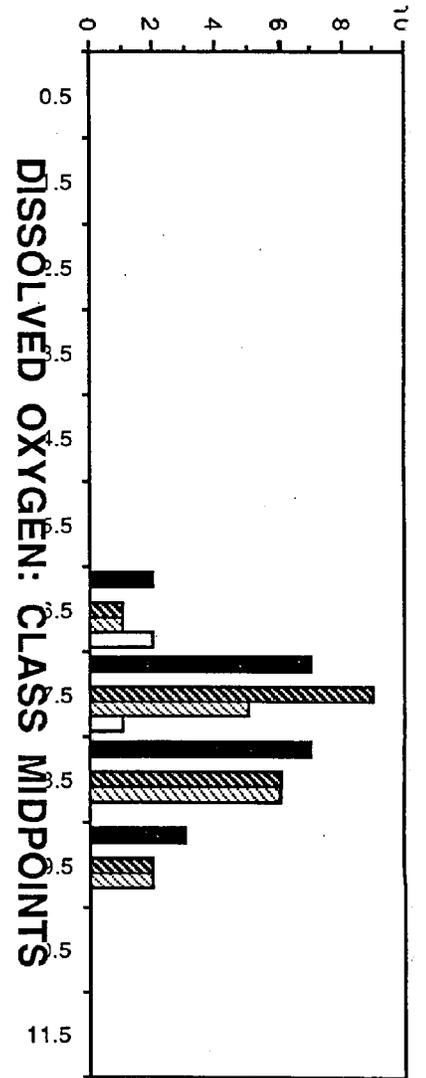


- Rhizosolenia delicatula
- ▨ Rhizosolenia stolerfothii
- ▩ Skeletonema costatum
- ▧ Thalassiosira oestrupii
- Thalassionema nitzschioides

- Cocconeis scutellum
- ▨ Cocconeis centralis
- ▩ Cyclotella striata
- ▧ Falcula hyalina
- ▦ Navicula cf. armophila
- Pyrocystis sp. 1

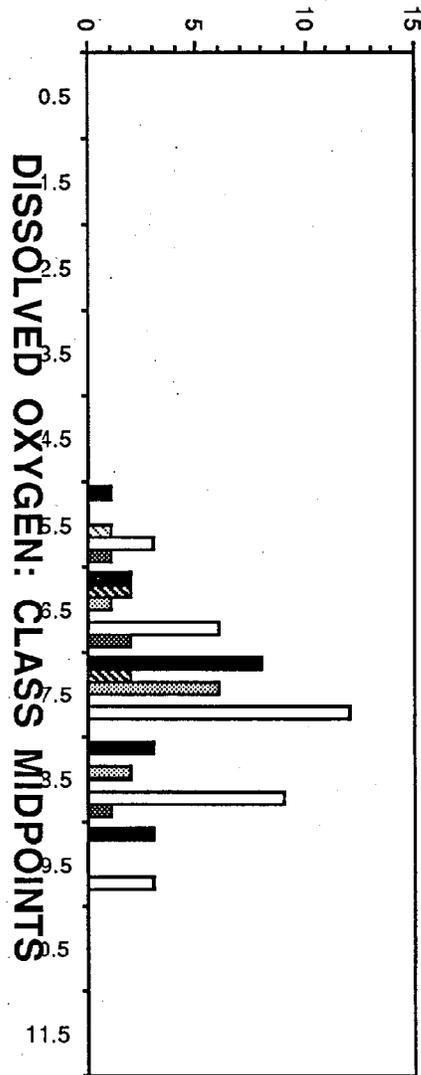
- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▩ Chaetoceros decipiens
- ▧ Chaetoceros didymus
- Chaetoceros socialis

OCCURRENCES



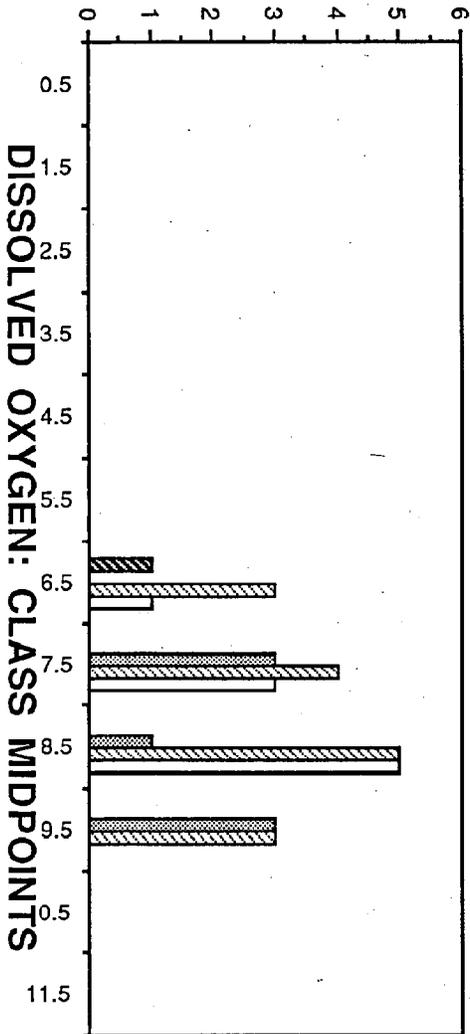
- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▧ Chaetoceros decipiens
- ▩ Chaetoceros didymus
- Chaetoceros socialis

OCCURRENCES



- Coscinodiscus centralis
- ▨ Falcula hyalina
- ▧ Rhizosolenia stollerfothii
- ▩ Cocconeis scutellum
- Cyclotella striata
- ▨ Navicula amorphila

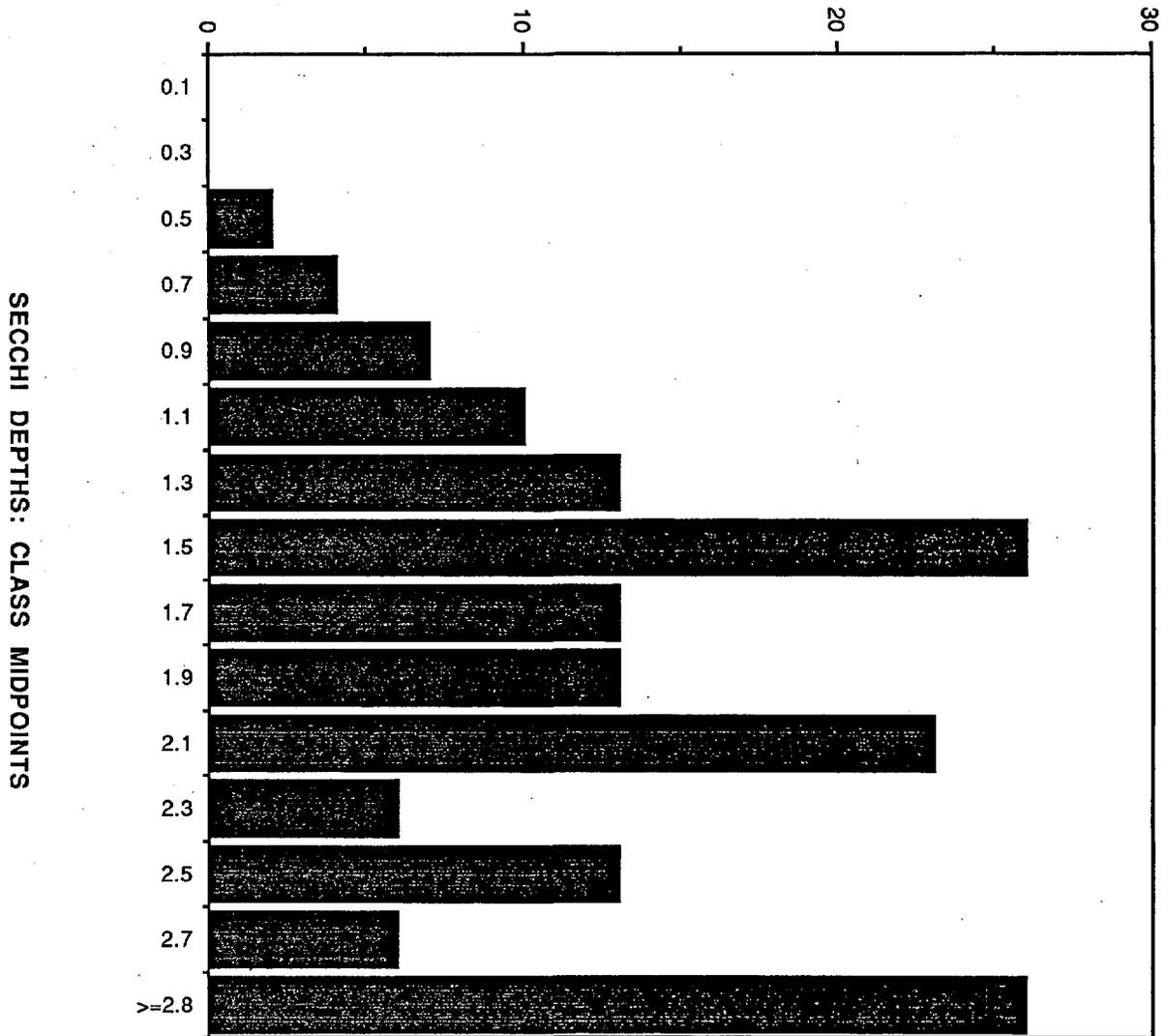
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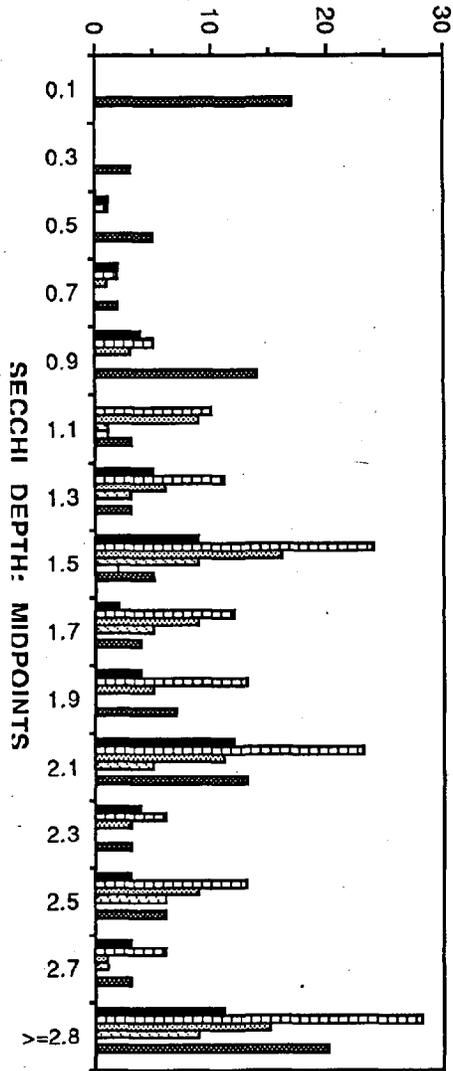
- Pyrocystis sp. 1
- ▨ Rhizosolenia delicatula
- ▧ Skeletonema costatum
- ▩ Thalassiosira oestrupii
- Thalassionema nitzechioides

OCCURRENCES

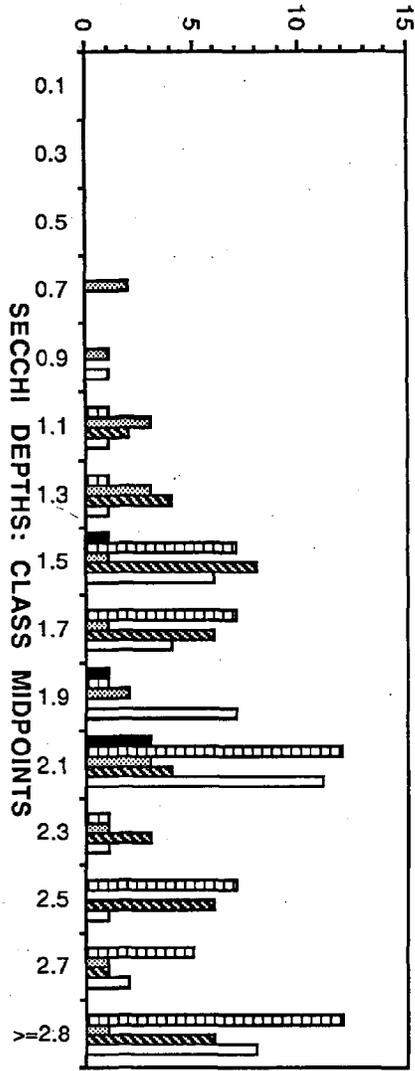
CHOCIAWHAICHEE BAY 64 MICRON-SAMPLES



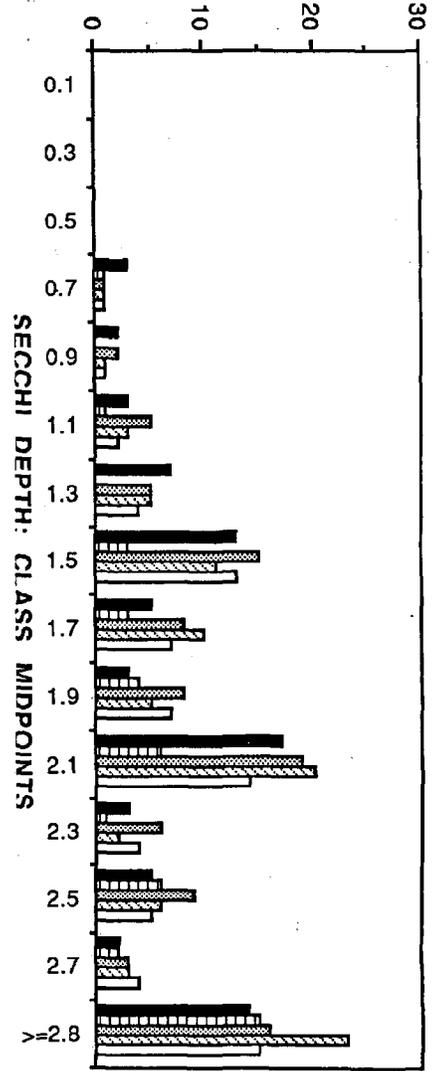
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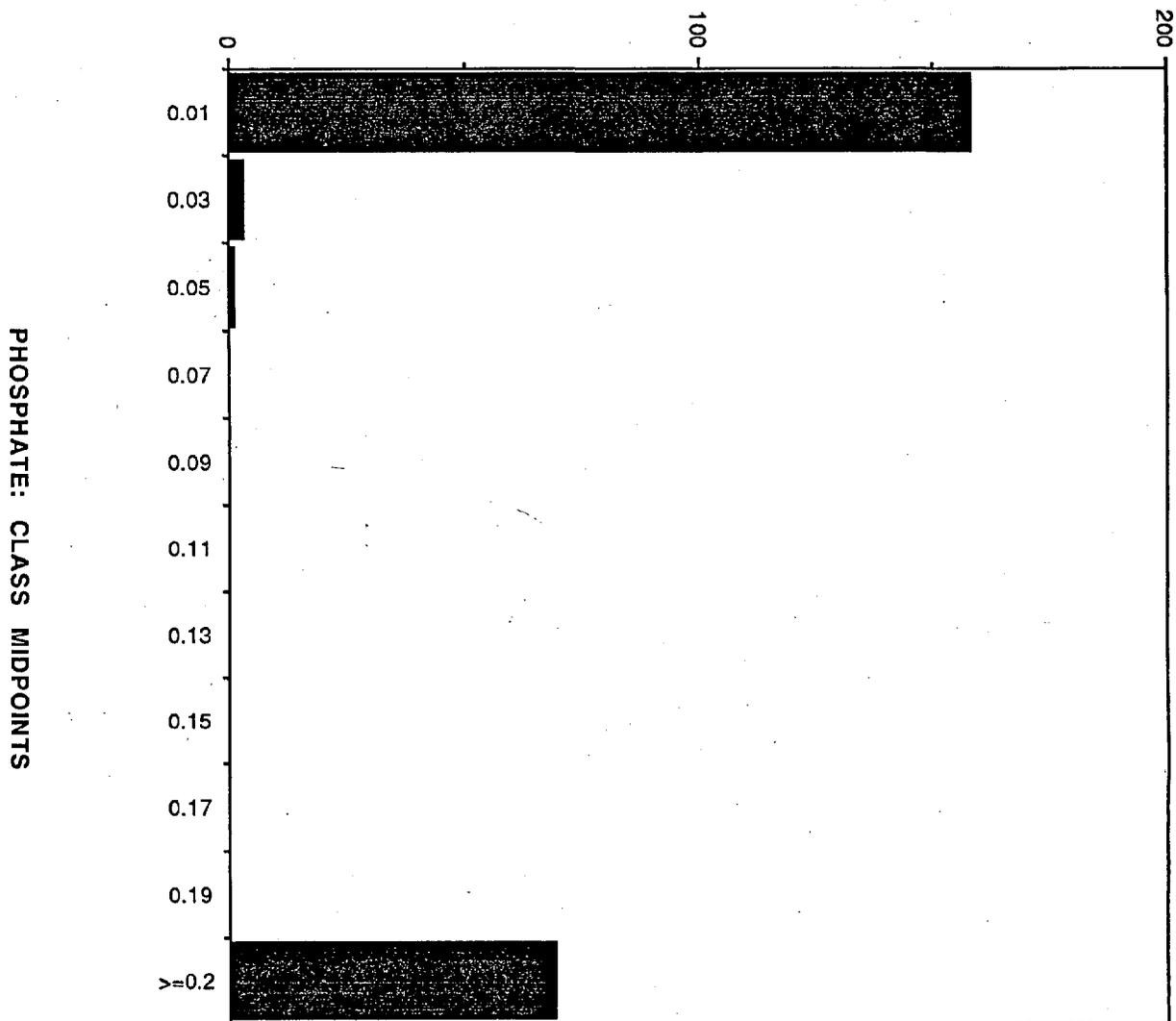
- Cocconeis scutellum
- ▨ Coscinodiscus centralis
- ▧ Cyclotella striata
- ▩ Falcula hyalina
- ▤ Navicula cf. armmophila
- Pyrocystis sp. 1

- Rhizosolenia delicatula
- ▨ Rhizosolenia stouterothii
- ▧ Skeletonema costatum
- ▩ Thalassiosira oestrupii
- ▤ Thalassionema nitzschioides

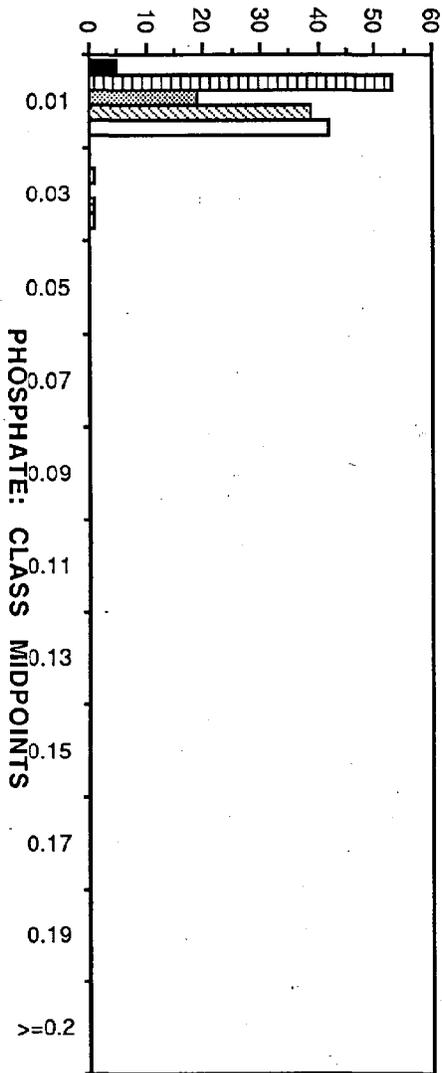
- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▧ Chaetoceros decipiens
- ▩ Chaetoceros didymus
- ▤ Chaetoceros socialis

OCCURRENCES

CHOCTAWHATCHEE BAY 64 MICRO-M SAMPLES

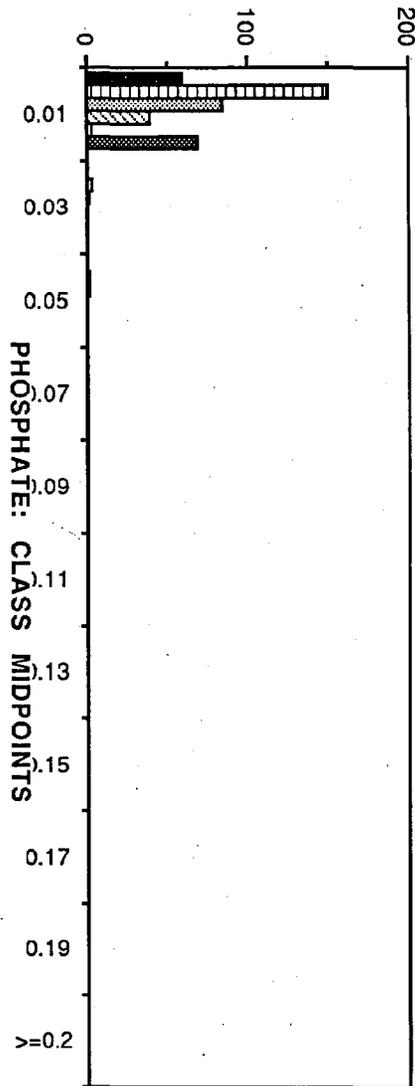


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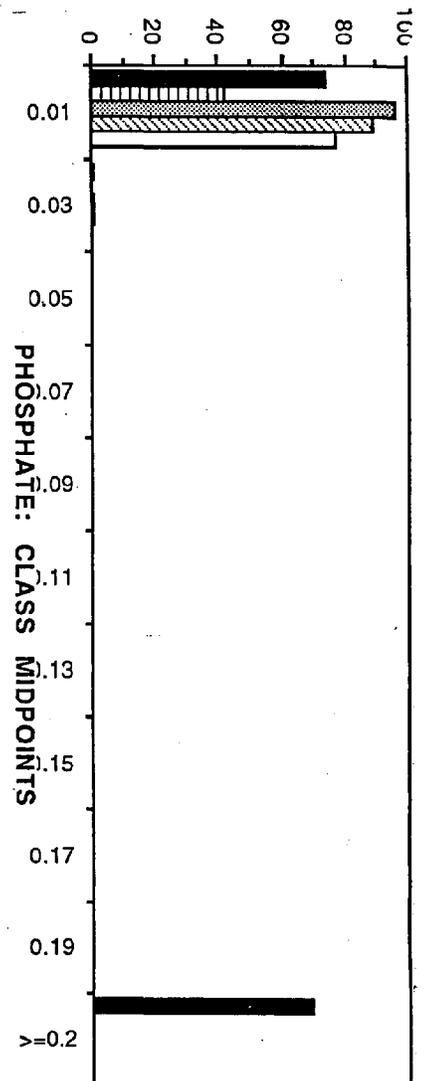
- Rhizosolenia delicatula
- ▨ Rhizosolenia stollerfothii
- ▩ Skeletonema costatum
- ▧ Thalassiosira oestrupii
- Thalassionema nitzschoides

OCCURRENCES



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- ▩ Cyclotella striata
- ▧ Falculla hyalina
- ▥ Navicula cf. amnophila
- ▦ Pyrocystis sp. 1

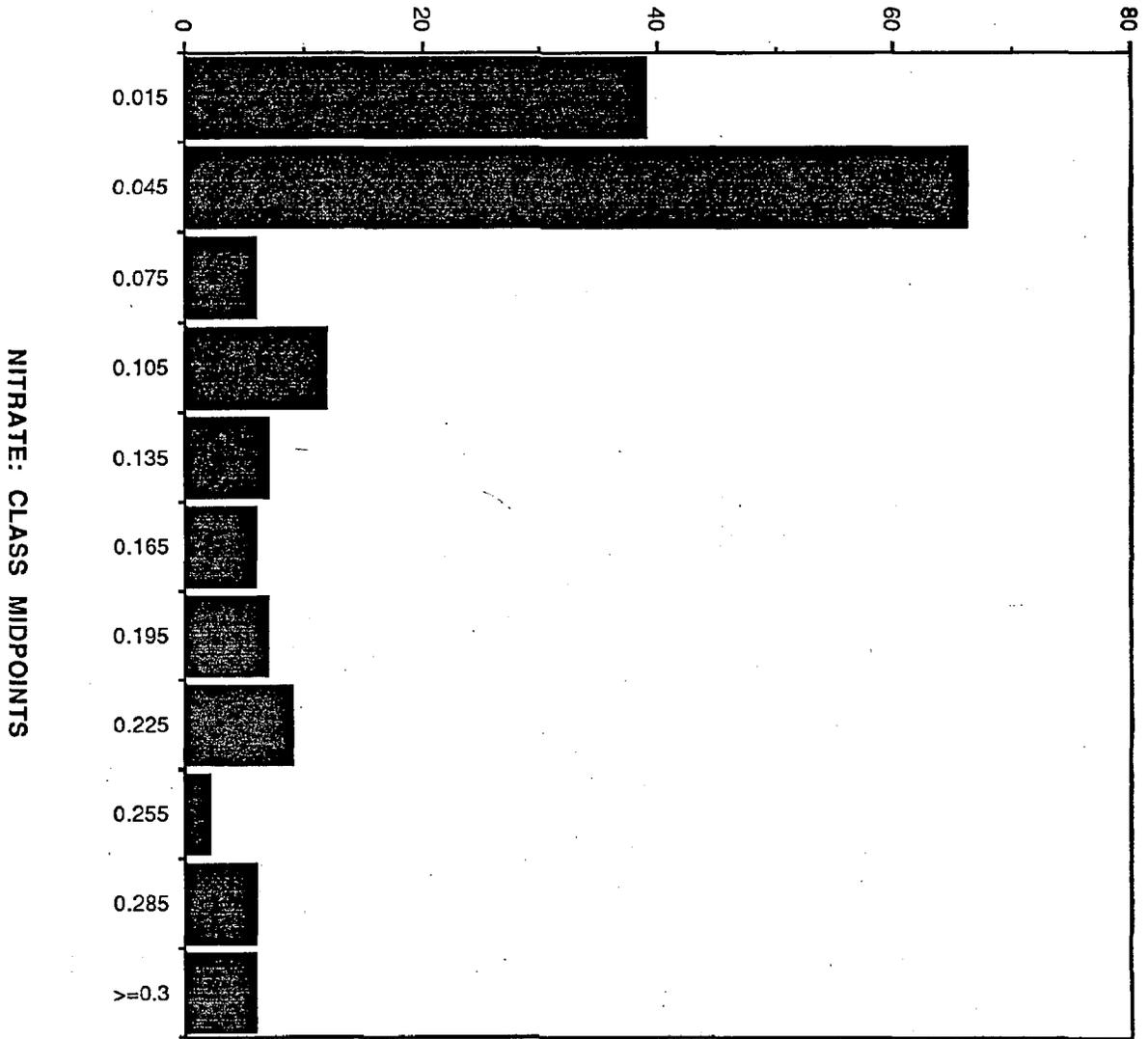
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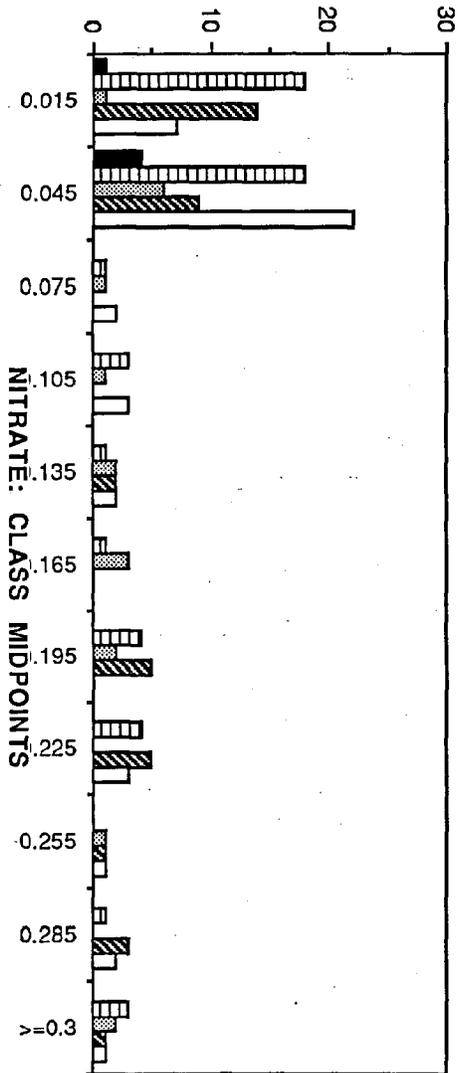
- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▩ Chaetoceros decipiens
- ▧ Chaetoceros didymus
- ▥ Chaetoceros socialis

CHOCTAWHATCHEE BAY 64 MICRO-M SAMPLES

OCCURRENCES

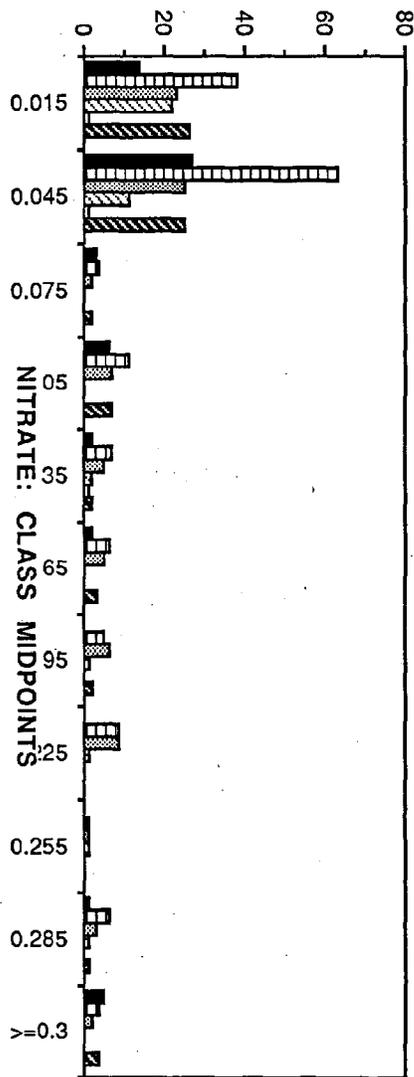


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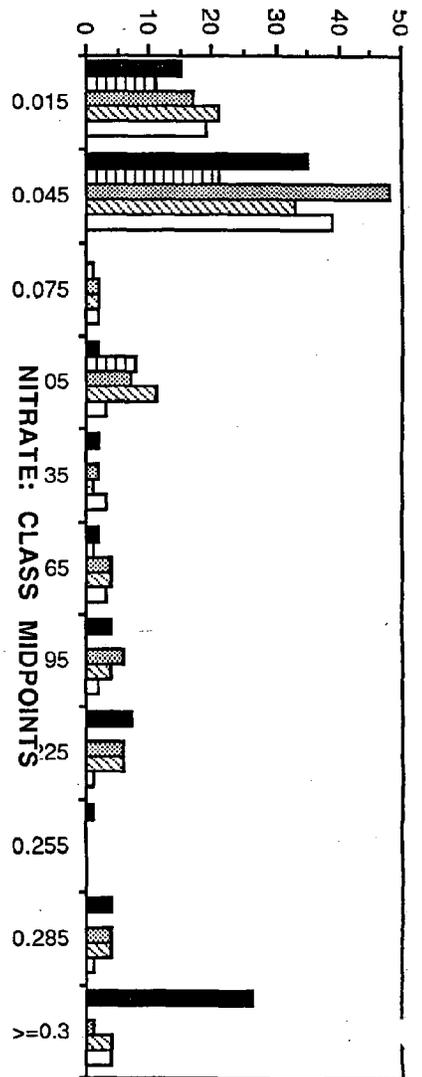
- Rhizosolenia delicatula
- ▨ Rhizosolenia stouterforthii
- ▩ Skeletonema costatum
- ▧ Thalassiosira oestrupii
- Thalassionema nitzschioides

OCCURRENCES



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- ▩ Cyclotella striata
- ▧ Falculla hyalina
- ▦ Navicula cf. ammophila
- Pyrocystis sp. 1

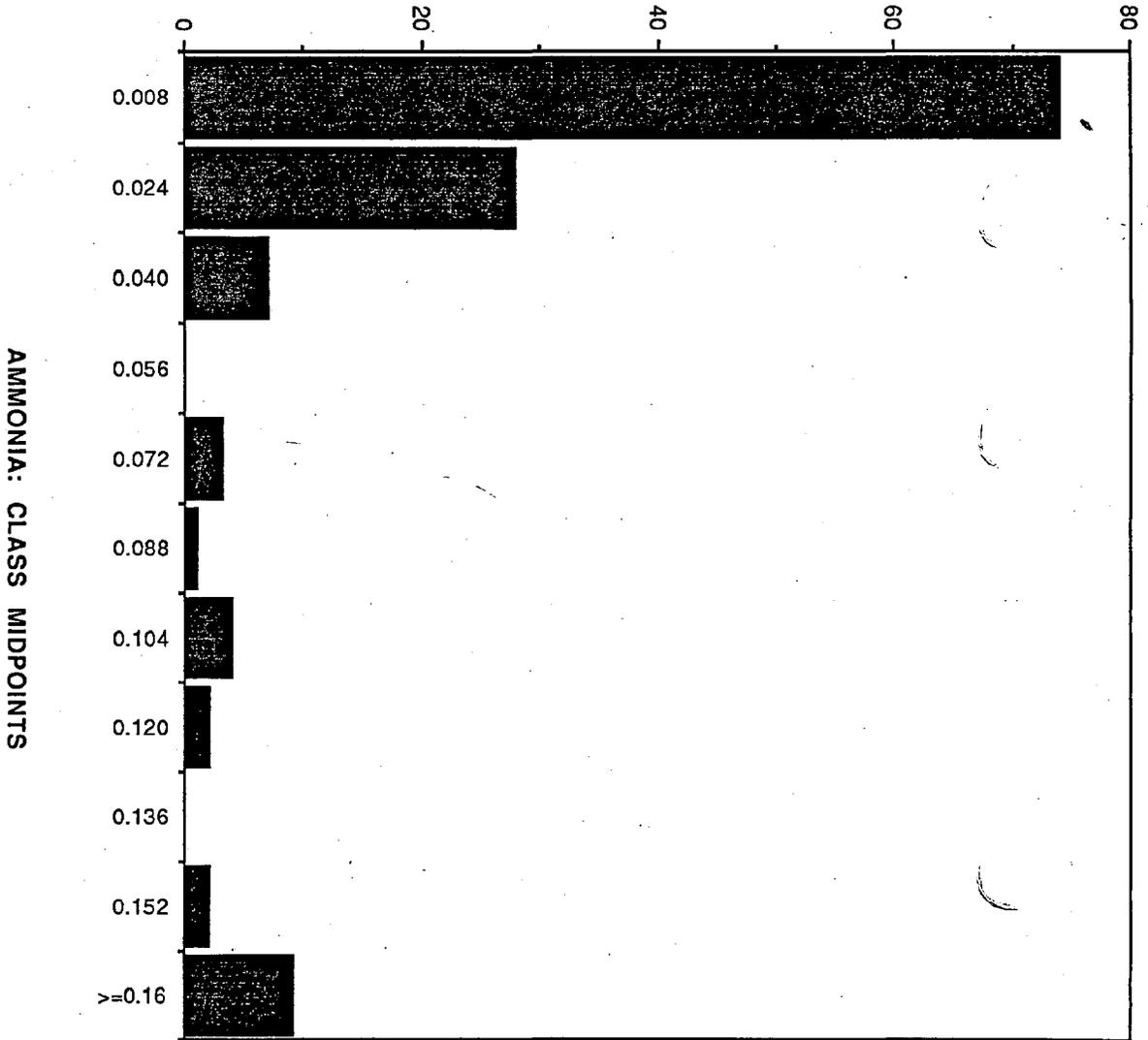
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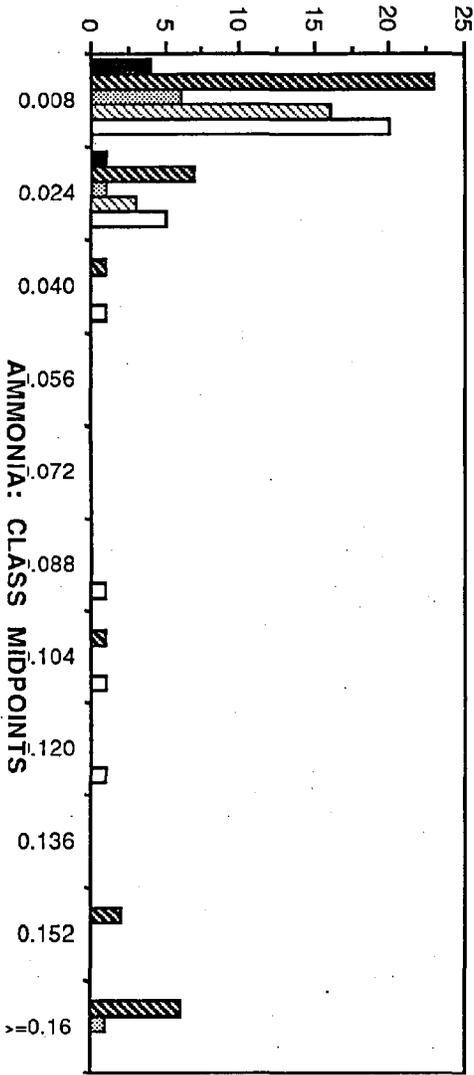
- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▩ Chaetoceros decipiens
- ▧ Chaetoceros didymus
- Chaetoceros socialis

OCCURRENCES

CHOCIAWHAICHEE BAY 64 MICHO-M SAMPLES

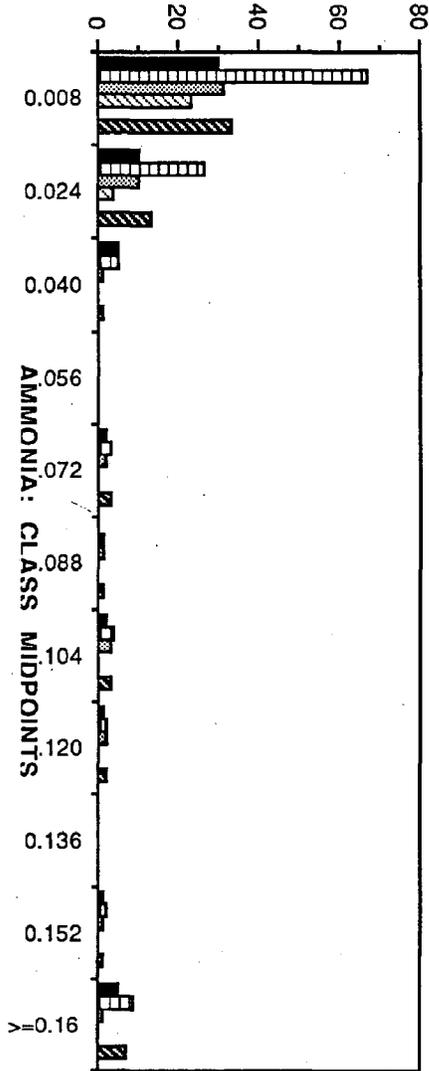


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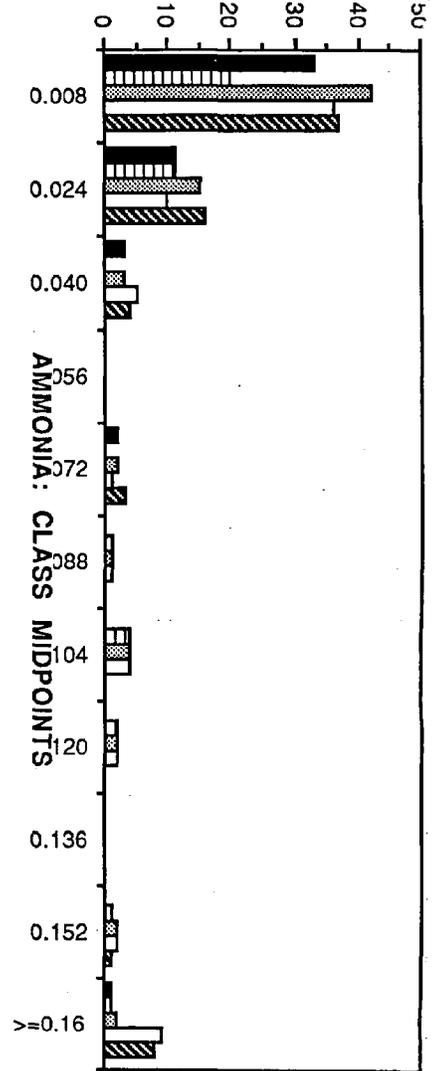
- Rhizosolenia delicatula
- ▨ Rhizosolenia stouterfothii
- ▩ Skeletonema costatum
- ▧ Thalassiosira oestrupii
- Thalassionema nitzschioides

OCCURRENCES



- Cocconeis scutellum
- ▨ Coscinodiscus centralis
- ▩ Cyclotella striata
- ▧ Falculla hyalina
- ▦ Navicula cf. ammoniphila
- Pyrocystis sp. 1

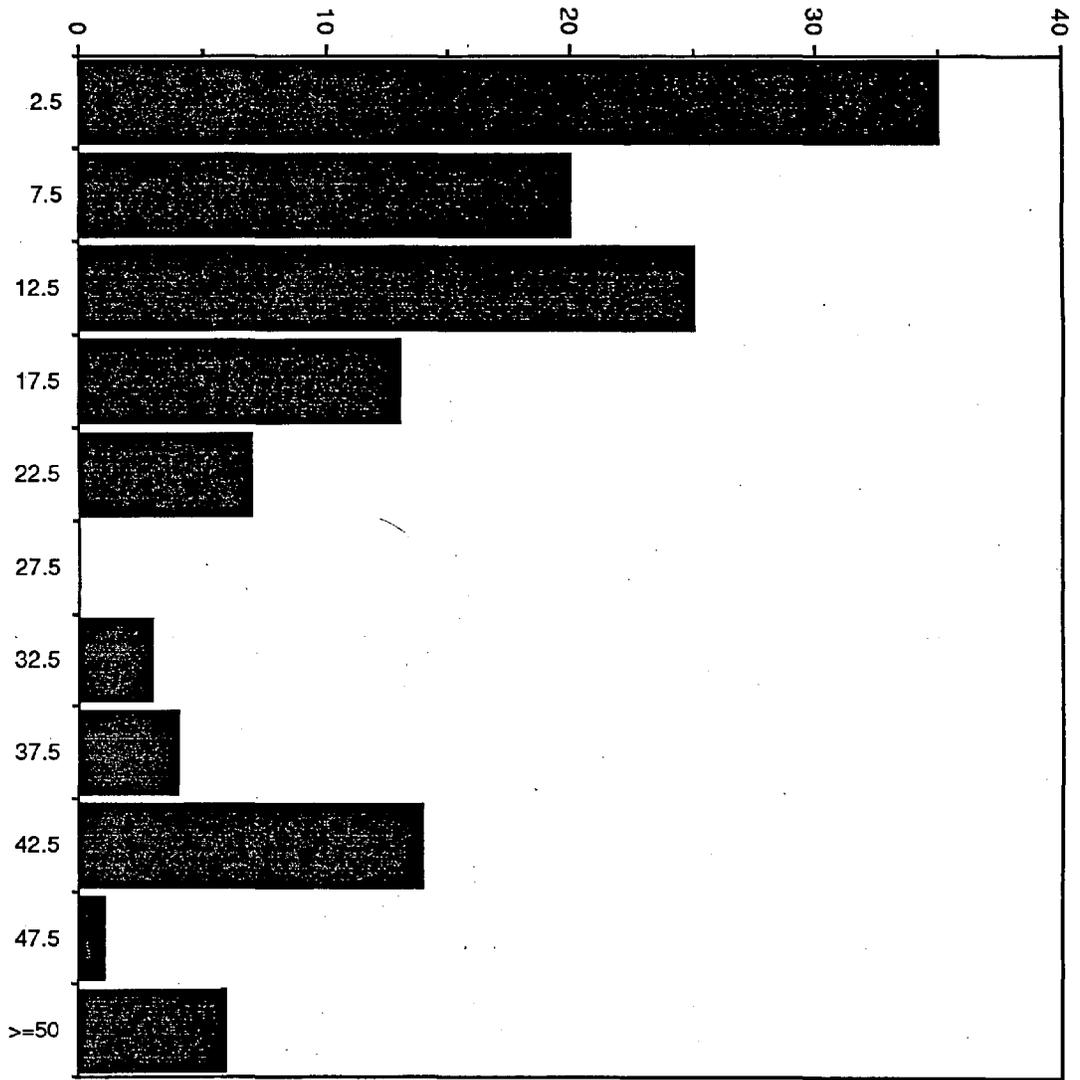
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- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▩ Chaetoceros decipiens
- ▧ Chaetoceros didymus
- ▦ Chaetoceros socialis

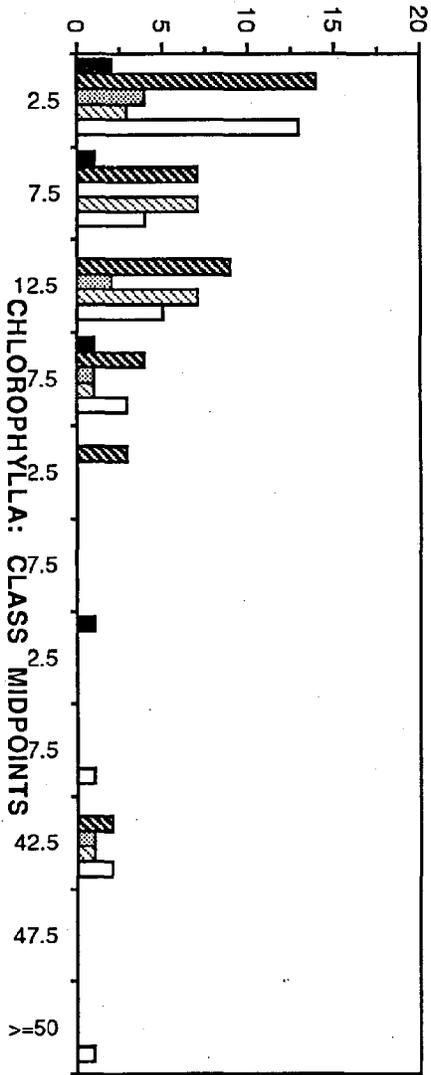
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CHLOROPHYLL A: CLASS MIDDPOINTS



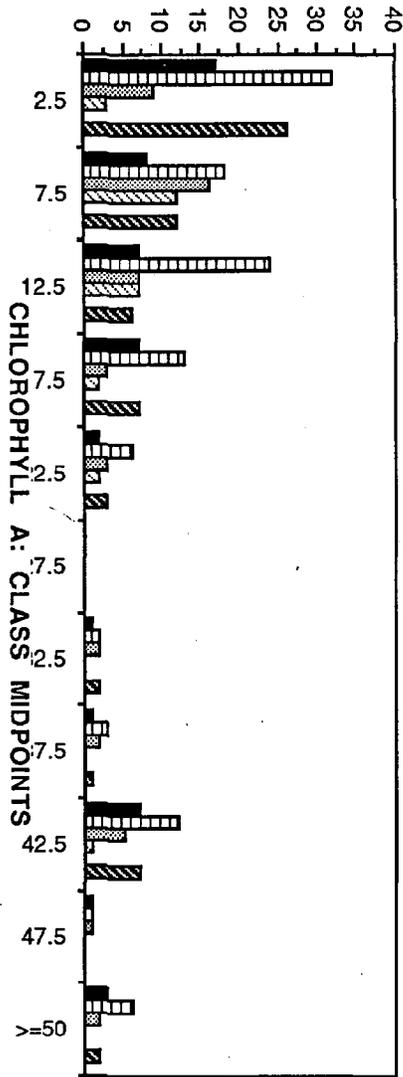
CHUOIAWTHAI ONEE BA) 64 MICROW-11 SAMPLES

OCCURRENCES



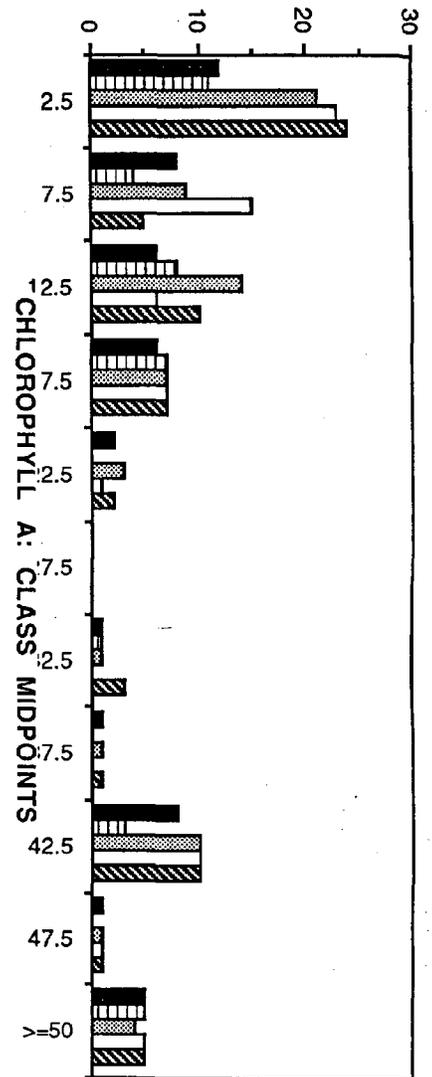
- Rhizosolenia delicatula
- ▨ Rhizosolenia stollerfothii
- ▩ Skeletonema costatum
- ▧ Thalassiosira oestrupii
- Thalassionema nitzschoides

OCCURRENCES



- Cocconeis scutellum
- ▨ Coscinodiscus centralis
- ▩ Cyclotella striata
- ▧ Falculla hyalina
- ▦ Navicula cf. ammophila
- Pyrocystis sp. 1

OCCURRENCES



- Chaetoceros brevis
- ▨ Chaetoceros coarctatus
- ▩ Chaetoceros decipiens
- ▧ Chaetoceros didymus
- ▦ Chaetoceros socialis

Secchi depth distributions (Figure 8) were ranged around 1.5 m. The species *Pyrocystis* sp. 1 was located in high numbers at relatively low levels of light penetration. Most of the other species were dominant at levels of relatively high light penetration according to the Secchi readings (usually > 1.5 m). The phosphate distributions were based on filtered samples; most such readings were very low so that the data, as presented, may not be representative of the response of phytoplankton to orthophosphate levels. The species *Chaetoceros brevis* may be an indicator of high phosphate levels. The data will be run with the unfiltered orthophosphate samples to verify this possibility. For now, these findings remain preliminary. Most of the species of phytoplankton were found at low levels of orthophosphate.

The nitrate distribution in the bay in terms of occurrence over the year-long study (Figure 8) was weighted around the low end of the spectrum. Once again, *Chaetoceros brevis* occurred at relatively high levels of nitrate which could be establish this species as an indicator of high dissolved nutrients. Other species such as *Thalassiosira oestrupii* and *Rhizosolenia stolterfothii* were also found in relatively high numbers at high concentrations of nitrate. Other phytoplankton species were clustered at the low end of the nutrient spectrum. Ammonia was also found to occur most frequently at the low end of the concentration spectrum. Species such as *Chaetoceros didymus*, *C. socialis*, and *Rhizosolenia stolterfothii* were indicators of high ammonia levels. Such species were also found at low concentrations of ammonia which means that they cannot be used as exclusive indicators of high ammonia levels. Indicators of chlorophyll a concentrations included most of the *Chaetoceros* species and *i*, *Coscinodiscus centralis*, and *Pyrocystis* sp. 1.

In most instances, the various phytoplankton species were found along relatively broad ranges of the various water quality distributions. This precludes

the use of any single indicator species as the sole representative of a particular habitat condition. However, when used in conjunction with the various community parameters, the phytoplankton as a group were highly indicative of various water quality conditions with particular emphasis on salinity and the nutrient distributions at culturally eutrophicated stations.

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APPENDICES

Appendix I: Sampling organization and protocols used during field operations in the Choctawhatchee River and Bay system and offshore areas of the Gulf of Mexico.

Appendix II: Quality Assurance and Quality Control protocols and Standard Operating Procedures used during the Choctawhatchee field programs.

Appendix III: Photographic atlas of dominant phytoplankton species found in the Choctawhatchee Bay system.

APPENDIX I

**CHOCTAWHATCHEE RIVER AND BAY SYSTEM
AND ASSOCIATED GULF AREAS**

**SAMPLING ORGANIZATION
AND PROTOCOLS**

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CHOCTAWHATCHEE RIVER STATIONS
FIELD SAMPLING PROGRAM

STA.	WAT.QUAL	NUTRIENTS	SEDIMENTS	PHYTOPLANKTON
* Guaged station				
60S (Pine Log Creek)	x	x	x	x
*61S (Choctaw- hatchee R.	x	x	x	x
62 (Seven Run Creek)	x	x	x	x
*63 (Choctaw- hatchee R. Discontinued 3/87)	x	x	x	x
64 (Bruce Cr.)	x	x	x	x
65S (Holmes Creek)	x	x	x	x
*66 (Choctaw- hatchee R. discontinued 3/87)	x	x	x	x
67S (Sandy Cr.)	x	x	x	x
*68S (Choctaw- hatchee R.)	x	x	x	x
69S (Wright's Creek)	x	x	x	x
*70S (Choctaw- hatchee R.)	x	x	x	x

**Station 68
Caryville**

Station 68 is located near Caryville, where US-90 crosses the Choctawhatchee River. The site is about 500 m in length and 150 m wide. Both banks are steep with much snag habitat. There are areas of overhanging willows on both upstream banks. The shoreline is sandy underneath the US-90 and railroad bridges. There is a granite breakwater located just north of the railroad bridge. The surrounding forest is primarily oak, gum, tupelo, and other floodplain species. There is no submergent or emergent vegetation. The water is usually turbid.

**Station 69
Wright's Creek**

Station 69 is located approximately 10 k northwest of Bonifay on SR-177. The site is about 75 m in length and 10 wide. The upstream area is narrower, has steep banks, and is surrounded by dense old growth forest. The banks are mostly snaggy habitat with several large trees lying in midchannel. The downstream right side is characterized by a large limestone outcropping and sand-clay beaches. There is a large cove where the water flow is usually slow. The right bank is steep with much scrub. The channel narrows considerably at the downstream end of the station and forms a shallow, pebbly riffle area with accelerated water flow. The area surrounding the downstream portions of the station is mostly flat and grassy. There is no emergent or submergent vegetation. The water is usually clear. Brook salamanders (*Eurycea*) were common.

**Station 70
Pittman**

Station 70 is located approximately 5 k west of Pittman on SR-2. The station is about 500 k in length and 150 m wide. The banks on both sides are very steep and high (bluff-like), and covered with old growth and floodplain forests. The upstream left side is mostly snag habitat with several large submerged trees. The downstream left side has a shallow cove with sandy and vegetated (maidencane) beaches, and a large stretch of overhanging willows. The upstream right side is mostly scrub and snag habitat. Midstream on the right side is an extensive limestone outcropping. The extreme downstream right side of the channel is snag habitat. The channel along the right side is very deep. The channel is shallow and sandy, with numerous sand bars, on the upstream half of the station. The water is usually turbid.

Station 64
Bruce Creek

Station 64 is approximately 3 k downstream from SR-81 (which is about 8 k south of I-10). The site is about 75 m in length and 10 m wide. This station is characterized by numerous large fallen trees and extensive snag habitat along both banks. The channel is deep except in a clay and pebble riffle area at the downstream end. The left bank is steep and primarily clay. The right bank is steep at the upstream and downstream ends and gently sloping in the middle. There is much clay and organic debris on the right bank. The forests surrounding this tributary are primarily oak, magnolia, and pine, with some scrub (primarily cabbage palm). The entire channel is overhung by a fairly thick canopy. There is no submergent or emergent vegetation at this station. The water is usually clear.

Station 65
Holmes Creek

Station 65 is located at SR-79 near Vernon. The site is approximately 500 m long and 25 m wide. The sides are mostly snag habitats. There are many large fallen trees and submerged pier pilings. The left side is primarily deep, with many complex root habitats, and patches of emergent and submergent vegetation. There is a large cypress-bordered cove on the left side. The right side is also deep, with steep limestone banks, and sloping forested banks. The forests on both sides of the site are primarily old growth hardwoods with thick overhanging canopies in places. The middle of the channel is mostly deep sandy bottom, interspersed with several large sand bar areas. These shallow areas are covered with submergent vegetation. The water is usually clear.

Station 67
Sandy Creek

Station 67 is located behind the I-10 and SR-81 rest area. The site is about 75 m in length and 15 m wide. It is mostly shallow, sandy riffle area. Some deep areas have developed (via scouring) in the channel in the proximity of large fallen trees. Both banks are mostly snag habitat, with large patches of scrubby and sandy areas on the left side. The banks are moderately steep and sandy. There is a large patch of submergent vegetation (*Ludwigia*, *Egeria* and *Nasturtium*) near the left bank. The surrounding forest is mostly old growth hardwoods and pine. The water is usually clear. A wild goat was observed.

Station 60 Pine Log Creek

Station 60 is located approximately 3 k south of Ebro on SR-79. The sample site is about 75 m in length and 10 m wide. The upstream portion of the site is bounded by thick strands of young pine and scrub oak. The upstream banks are characterized by a mixture of snags, roots, and scrubby bushes. The downstream banks are covered with scrubs and the surrounding shorelines are mostly grass. Both banks are steep. There is no submergent or emergent vegetation at this site. The middle of the channel is primarily shallow, sandy riffle areas with deep spots at both ends of the station. The right bank is clay underneath the SR-79 bridge. The water is usually darkly stained. Mice (*Peromyscus*), water snakes (*Nerodia*), and skinks (*Eumeces*) were commonly found along the scrubby shoreline.

Station 61 Ebro

Station 61 is located approximately 3 k west of Ebro on SR-20. The area sampled is about 500 m long and 150 m wide. Both sides of the channel are mostly high, steep banks (bluff-like). However, there are sandy (left side) and forested areas (right side) of low, gently sloping shoreline (downstream of the SR-20 bridge). The forests surrounding the river at this site are comprised of oaks and pine (left side, downstream); cypress and bay (inside fish camp channel); willows (portions of the left and right sides, upstream); and bay, tupelo, river hickory and oak mixtures (upstream, right). The right shoreline is predominantly snag habitat (snags, roots, and large fallen trees). There is a small strand of overhanging willows on the upstream right side. The left shoreline is characterized by large stretches of sandy beach (midstream and downstream), a large area of overhanging willows (upstream), and large areas of snag habitat (upstream and downstream). There is no submergent or emergent vegetation. The water is usually turbid. Water snakes and alligators (*Alligator mississippiensis*) were observed at this station.

Station 62 Seven Runs Creek

Station 62 is approximately 24 k south of I-10 on SR-81. The site is approximately 75 m in length and 10 m wide. The left side and upstream right side are bordered by thick strands of young oak. The far upstream portion of the station has a thick overhanging canopy. The left side of the channel is gently sloping and has large submerged leaf banks and patches of emergent vegetation (maidencane). Both sides have patches of submergent grasses (*Vallisneria* and *Sagittaria*) downstream. The right side of the channel is steep and comprised mostly of dense scrub and snag habitat. There is an old concrete pier in the middle of the channel. The water varies from clear to moderately stained. Alabama waterdogs (*Necturus alabamensis*), Amphiuma (*Amphiuma*), and three-lined salamanders (*Eurycea longicauda guttolineata*) were frequently observed in the leaf banks.

CHOCTAWHATCHEE BAY SYSTEM
FIELD SAMPLING PROGRAM

A. Physical-chemical Analysis

1. Basic water quality and nutrients

temperature	nitrate
pH	nitrite
dissolved oxygen	total Kjeldahl nitrogen
color	ammonia
turbidity	orthophosphate
secchi depth	total phosphorous
salinity	chlorophyll (a,b,c)
	particulate organic matter (POM) and carbon (POC)

The above parameters will be evaluated monthly from surface and bottom water samples at each of the 46 stations. In addition, chemical oxygen demand and coliform bacteria (fecal, total) will be determined for the river and bayou sites. The 24 hour surveys will include measurements of temperature, salinity, and dissolved oxygen. Such observations will be made at 4 hour intervals and at 1 meter depth increments.

2. Sediments

Basic data, including particle size distribution and percent organics, is to be taken quarterly at all 46 stations. Auxiliary information will include depth profiles of Eh, pH, dissolved oxygen, salinity, temperature, and nutrients. Spatial and temporal scope of this additional work is yet to be determined. We have an agreement with the Florida Department of Environmental Regulation that quantitative metal analyses will be provided for three of our stations.

B. Plankton

- ichthyoplankton (505 micron mesh net)
- zooplankton (202 micron)
- meroplankton (80 micron)
- phytoplankton (64 micron)

Surface samples will be obtained during the day in duplicate nets at 10 stations each month for each net type. Samples will be taken at the mid-bay station of each of the 10 transects and a set of data will be taken in Old Pass Lagoon (Station 36). At the 24 hour study sites both day and night samples will be obtained, where samples will be taken at the surface (nets), mid-depth (pump), and bottom (pump).

C. Fishes and invertebrates

Epibenthic fishes and invertebrates--seven two minute trawl tows monthly at all transect stations (32 sites). Two two-minute trawl tows will be taken at selected bayou stations and the Choctawhatchee River station.

Infaunal macroinvertebrates--10 cores (3" diameter) monthly at all transect stations (32 sites) and the Choctawhatchee River Station. Cores will be processed through a 500 um mesh sieve.

Additional fish samples--trammel nets, gill nets, and lift net samples will be obtained at five 24 hour stations each month. Exact net deployment (location, duration, etc.) will be at discretion of chief scientist, Dr. Christopher C. Koenig. Fishes too large to be returned to the laboratory will be identified and measured in the field. Stomachs will be removed and preserved and selected specimens will also have gonads removed.

D. Additional studies

1. seagrasses- A review will be made of the historic changes of emergent and submergent vegetation in the Choctawhatchee Bay system. This will be carried out largely with aerial photographs. By spring, 1986,

an experimental program will be established (based on the background data and historical reviews) which will include transplant experiments with seagrasses in an effort to enhance the productivity of this area. Data will be generated concerning standing crop biomass and productivity of existing beds with an emphasis on stations 26, 26A, 35, 37, 39, and 42.

2. Oysters- stations 16 and 17 will be analyzed for various factors associated with oyster propagation in the estuary. These data will be compared with ongoing studies by the Florida Department of Natural Resources.

All field work will be carried out using a fleet of boats from Florida State University with an estimated sampling time of 5-7 days. Such sampling will be performed during the third week of each month (weather permitting) over a 12 month period starting in September, 1985. Data generated will be placed in computer files for analysis with CARRMA'S software system and computers located in the Florida State University Computing Center. At the same time, a modeling program will be developed in conjunction with water quality models developed by the Northwest Florida Water Management District. As soon as all data have been entered (October, 1986), a series of model runs will be made using different management scenarios with application to predictions of the response of the Choctawhatchee Bay system to proposed structural modifications.

II. SAMPLING SUMMARY BY STATION

WQ--basic water quality and nutrients
 24-H--24 hour vertical profiles (temperature, salinity, DO)
 CC--chemical oxygen demand and coliforms
 PKT--plankton samples (D=day only, D/N= day and night)
 7TR--seven trawl tows, epibenthic fishes and invertebrates
 2TR--two trawl tows
 CORE--benthos (core samples), infaunal macroinvertebrates
 AUX NETS--auxiliary nets (trammel, seine, gill, lift)
 SED--sediments
 SG--seagrasses
 OYS--oysters

Station	Data Type										
	WQ	24-H	CC	PKT	7TR	2TR	CORE	AUX NETS	SED	SG	OYS
1 (Choc. River)	X		X			X	X		X		
2	X				X		X		X		
3	X			D	X		X		X		
4	X				X		X		X		
5 (LeGrange bayou)	X		X			X			X		
6	X				X		X		X		
7 (night stop)	X	X		D/N	X		X	X	X		
8	X				X		X		X		
9 (Alaqua bayou)	X		X			X			X		
10	X				X		X		X		
11	X			D	X		X		X		
12	X				X		X		X		
13 (Basin bayou)	X		X			X			X		
14	X				X		X		X		
15 (night stop)	X	X		D/N	X		X	X	X		
16	X				X		X		X		X
17 (Hogtown bayou)	X		X			X			X		X
18	X				X		X		X		
19	X			D	X		X		X		

	WQ	24-H	CC	PKT	7TR	2TR	CORE	AUX NETS	SED	SG	OYS
20	X				X		X		X		
21	X				X		X		X		
22	X			D	X		X		X		
23	X				X		X		X		
24	X				X		X		X		
24A	X				X		X		X		
25 (night stop)	X	X		D/N	X		X	X	X		
26	X				X		X		X	X	
26A	X				X		X		X	X	
27 (Rocky bayou)	X		X			X			X		
28	X								X		
29 (Boggy bayou)	X		X			X			X		
30	X				X		X		X		
31 (night stop)	X	X		D/N	X		X	X	X		
32	X				X		X		X		
33	X				X		X		X		
34	X			D	X		X		X		
35	X				X		X		X		
36 (Old Pass Lagoon)	X		X	D		X			X		
37	X				X		X		X	X	
38 (night stop)	X	X		D/N	X		X	X	X		
39	X				X		X		X	X	
40 (Garnier bayou)	X		X			X			X		
41 (Cinco bayou)	X		X			X			X		
42	X								X	X	
43 (the narrows)	X		X						X		
44 (open Gulf)	X								X		

III. PROTOCOLS (FIELD SAMPLING AND ANALYSIS)

A. Physical-chemical parameters

1. Water Quality Data

Field meters and equipment will be used on site for:

Temperature
pH
D.O.
Secchi
Salinity
Chlorophyll--filter 10 L on site

Field efforts at each station will consist of taking eight samples, each to be preserved according to protocol. One of these samples will be a quarterly sediment sample. COD and Coliform samples will be taken at designated fresh water input points of the bay.

2. Protocol for Chemical Sampling Choctawhatchee Bay

Coliform bacteria will be collected in sterile plastic bags. A sterile plastic bag will be filled beneath the surface of the water; a sweeping motion will be used with the open end of the bag kept towards the sweep. For depth, bags will be filled directly from the Kemmerer bottles. Samples will be immediately placed on ice.

TKN samples will be taken and acidified at the rate of 0.8 ml H₂SO₄/L and kept at 4°C in 1 L glass containers. Containers will be washed out with sample water and then filled from Kemmerer sampler or directly from surface waters.

Total P, PO₄ Nitrate, ammonia samples will be taken in 1L polyethelene containers. Containers will be washed out with sample water and samples will be taken from Kemmerer bottles or directly from surface waters. These samples will be analysed or frozen immediately.

Nitrite will be sampled from Kemmerer bottles in 500 ml polyethelene containers. Containers will be washed out with sample water before sample is taken. Samples will then be analysed or frozen immediately.

COD, color and turbidity will be taken in 500 ml polyethelene containers that have been washed out with sample water and then placed immediately on ice.

POC and POM will be collected in 1 l polyethelene containers, after washing containers with sample water. They will then be analysed or frozen immediately.

Sediments will be taken quarterly in 1000 ml coring containers and kept at 4°C.

Chlorophyll will be taken by filtering 10 l of sample water and storing filters at -20°C or will be analysed immediately.

3. Chemistry Methods Choctawhatchee Bay

Nitrogen (ammonia)--Nesslerization, measurement spectrophotometric at 425nm.

Nitrogen (nitrate)--Reduction, diazotization, measurement spectrophotometric at 400 nm.

Nitrogen (nitrite)--Diazotization. Measurement spectrophotometric at 505 nm.

Phosphorous (total)--Acid hydrolysis, persulfate oxidation. Measurement spectrophotometric at 880 nm.

Ortho-phosphate--Amino-naphthol reduction. Measurement spectrophotometric at 880 nm.

Total Kjeldahl nitrogen--Hydroxide-thiosulfate reagent digestion. Measurement spectrophotometric at 400 nm-425 nm (0.4-5mg/L) or with a 5 cm light path at 450 nm-500 nm (5-60 ug/L).

Determination of Chlorophylls--Millipore filtering with addition of $MgCO_3$, extracted by acetone and spectrophotometric measurement at 750 nm, 664nm, and 630nm.

Determination of Particulate Organic Carbon--Filtering, then "wet washing" with dichromate and conc. sulfuric acid. Measurement spectrophotometric at 440nm.

Particulate Organic Matter (POM)- filter sample, dry for 24 hrs at 100°C, ash at 500°C for 1 hr.

Total and Fecal Coliform--Hach multiple-tube fermentation technique, EPA approved.

COD--EPA approved COD reactor, premixed reagents, with concentration measured by spectrophotometric means.

*All procedures EPA approved or adapted from Standard Methods for the Examination of Water and Wastewater, Methods for Chemical Analysis of Water and Wastes (American Public Health Association, APHA) and a Manual of Chemical and Biological Methods for Seawater Analysis (Pergamon Press, 1984).

B. Protocol for Plankton Sampling

General Procedures:

Attach nets to frame securely

Set flowmeter setting to Zero or record initial flowmeter reading.

Check cod ends to make sure they are securely attached.

Launch nets.

Tow for specified time period at towing speed of 1-1.5 m/sec (2-3 knots).

Retrieve nets.

Record time of towing, boatspeed, flowmeter reading, time of day, tidal stage, depth, kind of tow.

Wash sides of net, then remove cod end, pour sample into prelabeled bottle, pour preservative (Approximately 1 : 100), seal, and store.

Specific procedures

Phytoplankton Sampling- Nannoplankton

Using Kemmerer Bottle obtain 500 ml of water/sample.

Preserve with 5 ml Lugol's Solution.

Phytoplankton Sampling - Net plankton

Tow 60 um net for 2 min (less if net is obviously no longer filtering water)

Preserve with 5 ml Lugol's Solution

Meroplankton Sampling

Tow 80 um net for 2 min. (less if not filtering at end)

Preserve with 2-3 ml Buffered formalin.

Zooplankton Sampling:

Tow 202 um net for 10 min.

Preserve with 2-3 ml Buffered formalin.

Ichthyoplankton Sampling:

Tow 505 um net for 10 min.

Preserve with 2-3 ml Buffered formalin.

When all sampling for the day is done wash all nets down with freshwater.

This will help keep the nets in good condition- do not wait until the next day since the corrosive effects of the sea water will already be affecting the nets.

Keep all nets out of the way during other sampling and put away and out of the sun as soon as possible.

Plankton Pumping Protocol

1. Begin pumping water at desired depth (use hose/marked at .25 m increments)
2. Place graduated (in liters) bucket under hose nozzle and measure time to fill to obtain pumping rate.
3. Place hose above plankton net and pump for appropriate time (pumping rate x time = volume).
4. Remove hose, remove cod end of net and remove sample.
5. Replace cod end and pump again for 2nd sample.
6. Preserve sample with appropriate preservative.

Notes for calculation of sampling volumes for towed plankton samples:

$$[\text{volume} = d' \times N \times A]$$

- (in m³)

Where d' = calibration factor for flow meter. This factor is generally between .15 and .16. It should be listed on technical material from supplier. It can also be calculated if distance towed is known ($d' = D/N$)

N = Number of revolutions of flow meter

A = Area of the net mouth in meter²

$$A = r^2$$

A 8" net = .0183 m² (r=.0762m r²=.0058m²)

A 20" net = .2033m² (r=.2540m r²=.0645m²)

Suggested volumes (EPA) 1.5-5m³

1m³=1000 liters

Plankton Towing Depth Calculation

Depth = L x cosine towing angle
Where L = length of line

C. Fish and Invertebrates

1. Protocol: Fish collection methods

- 1) Trawling: A standard 16 ft. otter trawl (try net) designed for biological sampling will be used to collect epibenthic fishes and macroinvertebrates. Seven 2 min. tows will be made at each designated station. Each tow should cover approximately 100 meters of bottom which means that tow speed should be about 1.85 mph. (3.0km/hr). Scope on the trawling line should be about 7:1; thus, for Choctawhatchee Bay, 210 ft. of line must be put out to trawl the stations 30 ft. deep.
- 2) Beach Seining: A 50 ft. 1/4 in. mesh beach seine will be used to collect near-shore fishes and invertebrates. Three consecutive 33 meter tows will be made with the long-shore current in areas conducive to this type of collecting (i.e., sandy beach areas) close to designated stations.
- 3) Trammel net collecting: A 100 meter trammel net (1 1/2 in. inside mesh, 12 in. outside mesh) will be used to collect bottom, midwater and surface dwelling fishes. The trammel net will not be fished conventionally. By the addition of leads and floats to the end thirds of the net, one third will fish midwater, and one third will fish the surface. The net will be set at designated stations at dusk and fished for about 4 hours. The net must be marked with flashing lights at night.
- 4) Night lighting: Fishes will be collected at night off the R/V Nectes by use of a 500 watt quartz flood light and several fishing methods: a) dipnet, b) lift net and c) small mesh gillnet. Dipnets will be operated by hand, lift-nets (1/8" mesh) will be suspended about 1 meter below the surface directly beneath the floodlight. The net will be lifted by hand when fishes have accumulated under the light. The sample mesh gillnets 1/2"-3/4" square mesh will be fished for four hours off the stern from 12 ft. poles suspended near the surface.

2. Coring: General protocol for macroinvertebrate coring

1. Take 10 cores (5.5cm diameter-10cm depth) per station.
2. Gently sieve each core through a 500 um screen with sea water.

3. Wash material retained on screen into 32 oz. plastic jar and add:
 - 1-2ml Rose Bengal solution*
 - 2-3ml Formaldehyde*
4. Double check labels and store sample.

*Since all samples are being sieved in the field, only small quantities of Rose Bengal and Formaldehyde will be necessary to insure adequate staining and fixation.

D. Trophic (Food web) analyses

Protocol: procedure for fish preservation and stomach content analysis

1. Fishes must be fixed in 10% buffered formalin (Sat. sodium borate-formalin).
 - a) fishes smaller than ca. 7cm SL can be preserved whole without opening body cavity.
 - b) fishes larger than 7 cm SL must be slit along the body cavity to expose the stomach to the formalin solution.
 - c) fishes too large to preserve should be weighed and measured (SL) and stomach and gonad removed and preserved in 10% buffered formalin.
 - d) preserved fish samples should be identified by waterproof labels inserted in the jars with the fish. Included on the label should be date, station number and method of collection. Written with pencil.
2. Fishes held in formalin for at least a week should be washed out in tapwater (allow several rinses of sufficient duration to remove formalin) and stored in either 50% isopropyl alcohol or 70% ethyl alcohol.
3. Weigh (gm) and measure (mm) fishes prior to removing stomachs. Use standard length (SL) on all bony fishes (straight line distance from tip of upper jaw to end of hypural plate). Measure total length (TL) of sharks and disc width of rays.
4. Remove stomachs and collect contents in vials with 70% ethyl alcohol and rose bengal.
 - a) pool stomach contents of smaller fishes of the same species and size class (CA. 10-15 stomachs depending on quantity of contents).
 - b) Determine stomach contents of larger fishes separately.
5. Transfer stomach contents to a series of nested sieves (2.0mm, 0.850mm, 0.425mm, 0.250mm, 0.125mm, 0.075mm) and wash contents through with an alcohol wash bottle or tap water.
6. Transfer each sieve fraction separately to small (CA. 2.5 in.) culture dishes (or watch glasses) and count particles of same size class and identify proportion of different organisms under a stereo microscope.

7. Transfer each identified size class to aluminum weighing dishes and dry at 60°C. After a constant dry wt. is reached weigh each fraction and record.
8. Calculate and record total dry wt. of stomach contents per fish and the dry wt. of each of the identified portions. (some classes may have to be identified as crustacean remains, shell fragments, etc.).
9. Lori Wolfe should be consulted for a printout of the identified stomach contents in 6 letter codes.

Chemical parameters (of water and sediments) analyzed
in the Choctawhatchee River-Bay project

<u>ORGANOCHLORINE PESTICIDES AND PCB'S</u> (sediments)		<u>CARBAMATES</u> (water)
Aldrin	Endrin aldehyde	Aldicarb
<u>a</u> -BHC	Heptachlor	Carbaryl
B-BHC	Heptachlor epoxide	Carbofuran
<u>w</u> -BHC	Kepone	Methomyl
q-BHC (lindane)	Methoxychlor	
Chlordane	Toxaphene	
4, 4' -DDD	PCB-1016	
4, 4' -DDE	PCB-1221	
4, 4' -DDT	PCB-1232	
Dieldrin	PCB-1242	
Endosulfan I	PCB-1248	
Endosulfan II	PCB-1254	
Endosulfan sulfate	PCB-1260	
Endrin	Mirex	
<u>ORGANOPHOSPHORUS PESTICIDES</u> (water)		<u>CHLORINATED FUNGICIDE</u> (water)
Azinphos methyl	Merphos	PCNB
Bolstar (Sulprofos)	Mevlnphos	
Chlorpyrifos	Monochrotophos	
Coumaphos	Naled	
Demeton	Parathion methyl	
Diazinon	Parathion	
Dichlorvos	Phorate	
Dimethoate	Ronnel	
Disulfoton	Stirophos (Tetrachlorvinphos)	
EPN	Sulfotepp	
Ethoprop	TEPP	
Fensulfothion	Tokuthion (Prothiofos)	
Fenthion	Trichloronate	
Malathion		
<u>METALS</u> (sediments)	<u>CHLORINATED HERBICIDES</u> (water)	<u>OTHER HERBICIDES</u> (water)
Aluminum	2, 4-D	Trifluralin
Arsenic	2, 4-DB	Benfluralin
Cadmium	2, 4, 5-T	Metribuzin
Copper	2, 4, 5-TP	Hexazinone
Chromium	Dalapon	
Iron	Dicamba	
Lead	Dichloroprop	
Mercury	Dinoseb	
Nickel	MCPA	
Zinc	MCPP	
	Atrazine	
	Alachlor	

OFFSHORE (GULF) STATIONS
OCTOBER, 1987; FEBRUARY, 1988

<u>station</u>	<u>Loran Reading</u>
M-1	29:51:82N, 84:26:77W
M-2	29:49:47N, 84:26:19W
M-3	29:49:30N, 84:28:14W
M-4	29:39:72N, 84:25:59W
E-1	29:57:72N, 84:00:37W
E-2	29:51:25N, 84:04:96W
E-3	29:47:62N, 84:06:44W
E-4	29:41:79N, 84:10:05W
A-1	29:36:48N, 84:57:54W
A-2	29:31:48N, 84:54:49W
A-3	29:26:75N, 84:51:43W
A-4	29:20:39N, 84:48:05W
C-1	30:20:20N, 86:32:50W
C-2	30:15:30N, 86:32:90W
C-3	30:08:50N, 86:35:01W
C-4	30:03:97N, 86:35:69W

APPENDIX II

**CHOCTAWHATCHEE RIVER AND BAY SYSTEM
AND ASSOCIATED GULF AREAS**

**Quality Assurance/Quality Control
and
Standard Operating Procedures**

**Robert J. Livingston
Center for Aquatic Research
and Resource Management
Florida State University
Tallahassee, Florida 32306**

**Quality Assurance Plan
for the
Choctawhatchee Project**

**Robert J. Livingston
Center for Aquatic Research and Resource Management
Florida State University
Tallahassee, Florida 32306**

Project Director

Associate Director

Consultant

Laboratory Director

Analyst

2. Contents

- 1) Title Page With Approval Signatures
- 2) Table of Contents
- 3) Project Description
- 4) Project Organization and Responsibility
- 5) QA Objectives for the measurement of Data
- 6) Sampling Procedures
- 7) Sample Custody
 - a) Field Sampling Operations
 - b) Laboratory Operations
- 8) Calibration Procedures and Frequency
- 9) Analytical Procedures
- 10) Data Reduction, Validation, and Reporting
- 11) Field and Laboratory Quality Control Checks
- 12) Performance and System Audits
- 13) Preventive Maintenance
- 14) Specific Routine Procedures Used to Assess Data Precision, Accuracy, and Completeness
- 15) Corrective Action
- 16) Quality Assurance Reports to Management
- 17) Personnel Qualifications, Resumes

3. Project Description

This project involves the monthly sampling of the water of the Choctawhatchee River and its' tributaries. There are three main stem stations: 61 (Ebro), 68 (Caryville), and 70 (Pittman). There are six tributary stations: 60 (Piney Log Creek), 62 (Seven Runs Creek), 64 (Bruce Creek), 65 (Holmes Creek), 67 (Sandy Creek), and 69 (Wrights Creek). A map of the stations can be found in appendix I.

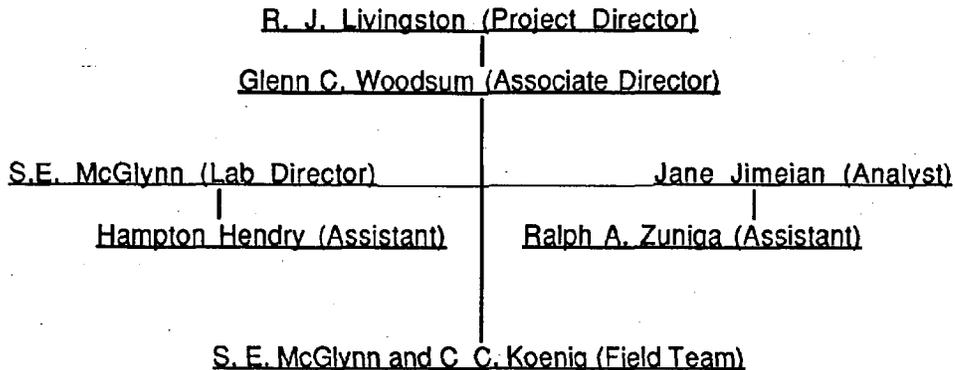
Conductivity, pH, temperature and dissolved oxygen will be determined in the field. Color, Turbidity, Biological Oxygen Demand, Chemical Oxygen Demand, Solids (suspended, dissolved, fixed and volatile), Particulate Organic Matter, Total Organic Carbon, Particulate Organic Carbon, Phosphate (dissolved, suspended, ortho and total), Nitrate, Nitrite, and Kjeldahl Nitrogen (total and organic).

Data will be used to develop a model of the Choctawhatchee River. The chemistry data will be combined with Hydrological data from the NFWFMD, and biological data from our biologists to complete the model

Phase One of the project went from December to January 1987, Phase Two of the project goes from December to January 1988. There may be a Phase Three of the project. In Phases One and Two 216 samples were taken. Nine per month for two years. These samples were taken in duplicate, so we actually took 432 regular monthly samples.

We also did Storm Monitoring, details of this program will be presented in Appendix 2.

4. Project Organization and responsibility



The Project Director is the ultimate source of responsibility in this project. The Project Director and Associate Director are responsible for all executive decisions dealing with personnel, equipment, sampling sites, sampling intervals, parameters monitored, methods, and final reports.

The Lab Director is responsible for implementing the policies of the Project Director, receiving samples, directing analysis, stocking the laboratory, maintaining equipment, updating the standard operating procedure, turning in all data to the analyst, and performing the more demanding analyses. The Lab Director is the Quality Assurance Officer.

The Analyst is responsible for all the raw data turned in by the Lab Director, computing the finished data, filing the finished data, and reviewing the finished data with the Laboratory Director.

The Field Team collects the samples in the field and ships them to the laboratory. They are responsible for the samples until they reach the laboratory, the Chain of Custody forms, and the equipment while they are in the field.

5. Quality Assurance Objectives

PARAMETER	METHOD*	MATRIX	PRECISION (%RSD)	ACCURACY (sd)	COMPLETENESS (%)
Color	204B	L	9	3	100
Conductivity	205	L	9	3	100
TDS	209B	L	12	50	100
TSS	209C	L	12	10	100
POM&PIM	209D	L	12	4	100
Salinity	210A	L	9	3	100
Temperature	212	L	5	2	100
Turbidity	214A	L	6	4	100
Alkalinity	403	L	3	.5	100
Ammonia	417B	L	6	.01	100
Nitrate	418C	L	7	.05	100
Nitrite	419	L	6	.001	100
TKN&OKN	420B	L	12	.2	100
D.O.	421F	L	4	.1	100
pH	423	L	5	.2	100
Phosphorus	424F	L	7	.005	100
TOC	505A	L	7	.1	100
BOD	507	L	7	.1	100
COD	508C	L	17	5	100
Chlorophyll	4.1†	L	18	NA	100

*Unless stated otherwise methods are from *Standard Methods for the Examination of Water and Wastewater*, 1985, 16th edition, APHA, AWWA, and WPCF.

† *A Manual of Chemical and Biological Methods of Seawater Analysis*, T. R. Parsons, Y. Maita, and C. Lalli, 1984, Pergamon Press.

6. Sampling Procedures

*"The result of any test can be no better than
the samples on which it is performed."*

An old Axiom

The objective of sampling is to collect a portion of material small enough in volume to be transported conveniently and handled in the laboratory while still accurately representing the material being sampled. This implies that the relative proportions or concentrations of all pertinent components will be the same in the samples as in the material being sampled and that the sample will be handled in such a way that no significant changes in composition will occur before the tests are made.

Sample bottles, usually half gallon plastic milk containers must be rinsed with the water being sampled at least three times. Sample containers that are to be re-used are washed withalconox™, rinsed at least three times with dl water, dried, and then sealed to avoid any contamination. If phosphates are to be analyzed the sample containers are acid washed with warm 10% HCl, and if trace metals are to be analyzed the sample containers must be acid washed with 10% nitric acid.

Samples collected at a particular time and place can represent only the composition of the source at that time and place. Grab samples are collected with a

Kemmerer sampler at the bottom of the water column. Composite samples are taken with an integrated sampler. Avoid collecting detritus by taking the sample a few centimeters above the soil/water interface. Surface samples are collected by lowering an inverted sample container beneath the water/air interface and righting it. Avoid collecting any flotsam and jetsam by filling the sample container 5cm beneath the surface. Avoid entrapping air in the filled sample container. The sample must be properly preserved until it is received at the laboratory.

When a source is known to vary with time, samples must be taken with appropriate frequency to monitor the extent of these variations. In such a situation, the location and the time of sample collection must be accurately duplicated. In open water, a Loran can assure site location to within a hundred feet, otherwise landmarks must be judiciously chosen.

Samples are put on ice in the dark immediately to assure stability of constituents until they can be analyzed in the lab. Before delivery of the sample to the lab, a chain of custody form must be filled out detailing the volume of the sample, the location of the site, the date and time of sampling, the name of the samplers, the project and/or the parameters to be analyzed, the technique by which the sample was obtained, and the methods of preservation. Completed chain of custody forms must be received and signed by our laboratory personnel. Then they are filed at the laboratory where they are a record of sample history. All subsequent tests performed on these samples are recorded in the our Laboratory Log Book.

Samples are to be delivered to the lab with all possible haste, if delivery time exceeds 6 hours, correct preservation techniques must be observed. IF these guidelines are observed samples on ice, with a proper chain of custody form will be accepted by the laboratory. Once received samples are allowed to rise to ambient temperature before analysis can begin.

The unequivocal preservation of samples is fundamentally impossible. Regardless of the preservation technique, complete stability for every constituent can never be achieved. It is best to analyze samples as soon as possible after collection, and then to judiciously determine the type of preservation to be utilized.

Measurement	Container	Preservative	Holding
Color	P, G	Cool, 4°C	48 hours
Conductance	P, G	Cool, 4°C	28 days
pH	P, G	None	None
Filterable Residue	P, G	Cool, 4°C	7 days
Non-filterable Residue	P, G	Cool, 4°C	7 days
Temperature	P, G	None	None
Turbidity	P, G	Cool, 4°C	48 hours
Alkalinity	P, G	Cool, 4°C	14 days
Ammonia	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Kjeldahl Nitrogen	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Nitrate	P, G	Cool, 4°C	48 hours
Nitrite	P, G	cool, 4°C	48 hours
Oxygen, dissolved	P, G	None	None
Ortho-Phosphate	G	Cool, 4°C, no acid	48 hours
Total Phosphate	G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Phosphate, T. dissolved	G	Cool, 4°C, H ₂ SO ₄ to pH < 2	24 hours
BOD	P, G	Cool, 4°C	48 hours
COD	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Organic Carbon	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days

7. Sample Custody

Sample custody is important from a legal standpoint. We feel that it is especially important that the great care taken in their work should be documented. No time passes after the sample is taken without being documented. Chain of custody forms cover the sample in the field, in transit, and in the laboratory. While in the laboratory a log book charts the various analysis performed on the sample. After all the experimentation is complete the Chemist performing the analysis initials the original Data Sheet. The original Data Sheets are handed to the Analyst by the Laboratory Director. The original Data Sheets are filed. The Data is punched into our Macintosh Computer System by our Analyst and her assistants. After the Data is punched it is checked by our Analyst who initials the original Data Sheet signifying that it has been checked. After calculations are performed on the Data it is stored on Bernoulli Disks.

a. Field Sampling Operations

Equipment that goes in the field is prepared prior to each field trip. All field meters are calibrated in the laboratory prior to each field trip. Typically, field crews need a Salinometer, a Dissolved Oxygen Meter, a pH meter, and a thermometer.

The salinometer we use in the field is a YSI SCT Meter. It is calibrated against a standard saline solution before each field trip. Usually if it does not calibrate the probe must be cleaned with an HCl/isopropanol solution. The temperature sensor is calibrated

against an NBS thermometer. These methods are detailed in the YSI Operators Manual (see SOP).

The dissolved oxygen meter we use in the field is a YSI DO Meter. It is Winkler Calibrated quarterly. Before each field trip it is calibrated against an oxygen saturated water solution. Before use in the field it is air calibrated. The DO Membrane is replaced prior to use. If response is sluggish, or if there is any difficulty in calibration, clean the silver electrode with a 14% Ammonium Hydroxide Solution, and clean the gold electrode with an abrasive rubber eraser. The temperature sensor is calibrated against an NBS thermometer. These methods are detailed in the YSI Operators Manual (see SOP).

The pH meter we use in the field is a Corning 610 pH Meter. Of all our field equipment this is the most prone to water damage. If it gets very wet the electronics will be damaged. The calomel electrodes are kept filled with a saturated KCl solution. If response gets sluggish clean the semipermeable membrane in warm pH 10 buffer, and look for air bubbles in the ceramic junction. Before use the meter is calibrated to three different pH buffers. The buffers are for pH 4, 7, and 10. These buffers are taken into the field, and the meter is calibrated prior to each reading in the appropriate pH range. These methods are detailed in the Corning Operators Manual (see SOP).

The meters, a graduated depth line, and the Secchi must be signed out to the field crew by the Laboratory Director, who makes sure all equipment is in full operating condition.

The Field Crew labels all sample bottles after they have been cleared by the Laboratory Director. The date, location, and station identification number must be written on every sample bottle with a permanent marker. The Field Crew obtains Field Data Sheets from the Analyst, it is the Analysts duty to work with the Project Director and specify sampling locations and frequency, that are clearly stated on the Field Data Sheets.

The Field Crew is responsible for custody of the sample until it is released to a carrier or the laboratory. The chain of custody form initiated by the Field Crew states the location, time, date, method of sampling, number of replicates, and method of preservation of each sample. The persons in charge of the sample until it reaches the laboratory must sign indicating the time when the sample came into their custody, and when they relinquished the sample to someone else.

b. Laboratory Operations

The Laboratory Director is the sample custodian at the laboratory facility. He signs for incoming samples and obtains documents of shipment. Once the samples arrive at the laboratory their progress through analysis is documented in the Laboratory Log. All analysis, new reagents, standard curves, etc. are signed and dated in this book by the Chemist.

The Laboratory Director is responsible for the analysis of the sample. Results of analysis are recorded on the serially numbered Laboratory Data Sheets. These are a complete record of the results of all the analysis performed on the samples from a given field trip. The Laboratory Data Sheets also record the standards which are run. The Laboratory Log details the observations and calibrations that may have occurred during the run. It provides an accurate record of the date and time of each run, and of the preservation status of any given sample.

8. Calibration Procedures and Frequency

Spectrophotometers

(Bausch & Lomb Spectronic 2000, Beckman DB/G,
and a Carl Zeisse Elco II)

A series of seven standards that bracket the range of concentrations anticipated for any given parameter are run with every analysis as specified in our SOP. A standard curve is generated from this data, and the slope and intercept are reported on the original data sheet. If the R^2 is less than 0.90 the spectrophotometer is serviced and the analysis is run again on a different instrument until a satisfactory calibration curve is obtained.

All the calibration curves are kept on file by the Laboratory Director. Each new Calibration Curve is compared by the Laboratory Director with previous Calibration Curves from the same parameter. If there are significant differences between the curves this is also cause for the analysis to be rerun.

All standards are made in our laboratory from ACS reagents according to EPA specifications. A Stock solution is made first. The flask is dated and signed by the Chemist who made it. A Standard solution is then made from the concentrated Stock Solution. The flask is dated and signed by the Chemist who made it. Standards run with the analysis are serial dilutions of the standard solution. They are made fresh for every analysis.

All Stock and Standard solutions are recorded in our Laboratory Log Book when they are made. Old standards are discarded as they go bad. All the standards are dated. We follow EPA guidelines in ascertaining their lifetime, and we check their age prior to use. We also do a visual inspection of the standard before use. The ultimate test of the standard is if it generates a good standard curve, however a bad standard curve can also be caused by malfunctioning equipment.

We can trace our standards by checking in our Laboratory Log for the current standard in use during any given analysis.

Mettler Balances

(Mettler H33, and a Mettler H15)

Our Mettler Balances are calibrated annually by certified technicians. If there are any problems with one of them we have a backup. With the two balances we can check the accuracy of one against the other.

Turbidometers

(2 Hach model 2100A)

These are calibrated with every use with commercial Hach standards as specified in our SOP.

Colorimeters

(Hach DR-A, and a Hach DR-3)

These are calibrated with every use with commercial Hach standards as specified in our SOP.

DO Meters

(3 YSI model 57)

These are air calibrated with every use. Winkler calibrated quarterly as specified in our SOP.

SCT Meters

(3 YSI Model 33)

These are calibrated prior to each use with a standard saline solution as specified in our SOP.

PH/mv Meters

(Beckman Century SS, and 3 Corning 610A)

These are calibrated with Buffered solutions obtained commercially at pH 4, 7 and 12. The mv scale used with an Ammonia sensitive Electrode is calibrated with the seven point standards used for the Ammonia test as specified in our SOP.

9. Analytical Procedures

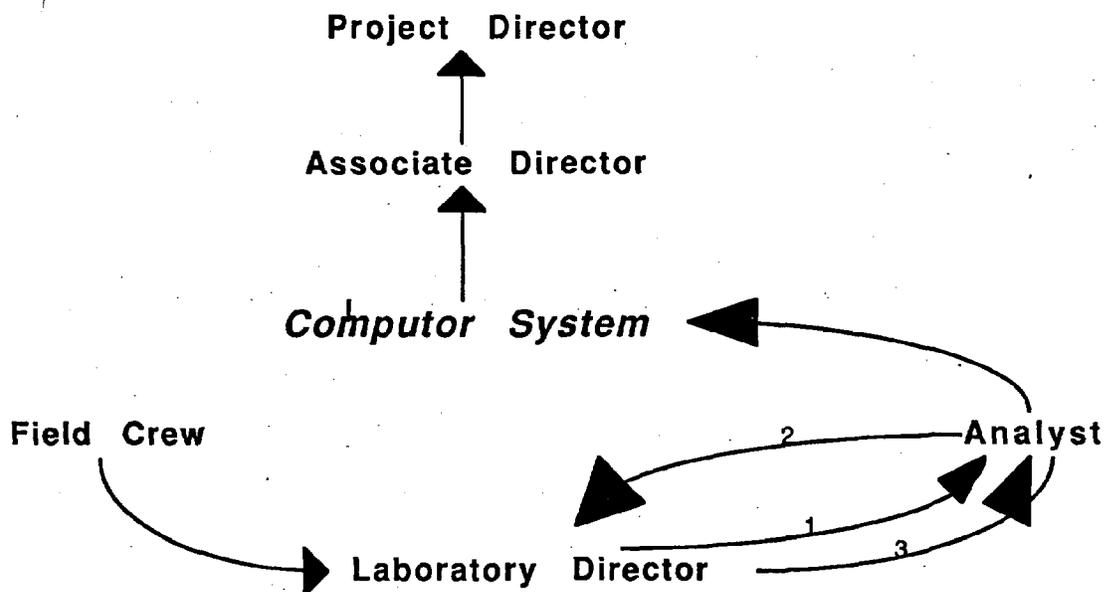
Only EPA approved procedures are used in our laboratories. All our laboratory procedures are detailed in our Center for Aquatic Research and Resource Management Standard Operating Procedures, (SOP).

10. Data Reduction, Validation, and Reporting

Only EPA approved procedures are used in our laboratories. All our formulas are detailed in our Center for Aquatic Research and Resource Management Standard Operating Procedures, (SOP).

All data is reported to our analyst. The Analyst and the Laboratory Director then preliminarily validate the data, which is then sent to the project manager for final validation. The criteria used to validate data integrity is adherence to our system of absolute standards, the conformation of replicates to the %RSD as stated in section 6.5, and the recovery of spiked samples from the field.

Data Flow



11. Field and Laboratory Quality Control Checks

A random approach is taken as the basis of this statistical audit of our sampling program. All sampling in the field is done in triplicate. From these triplicate samples our replicate analysis are performed.

We run 10% replicates on every analysis performed in our laboratory. Replicates are reported on the Data Sheets turned in to the Analyst. It is left to the discretion of the Chemist to perform replicates within the necessary 10% framework. We expect all replicates to correspond to our Quality Assurance Objectives detailed in section 5. If they do not corrective action is outlined in section 15.

Every sampling we undertake gets at least one spiked field sample for each parameter analyzed. We run a field blank along with our field spikes. If a project continues over a period of time spiking is repeated with each sampling mission. Field Spikes are usually performed on the natural water being tested. If there is any particular quality of the water that would interfere with our analysis this should help make it apparent. The natural value of this parameter will have to be analyzed in our lab and then subtracted from the spike. We expect 95% recovery of all spikes. The results of our Field Spikes are kept on file by the Quality Assurance Officer, and are reported in our quarterly Quality Control Report to the Project Director.

Equipment Blanks and Calibration Standards are employed with every run of our laboratory equipment. From the results a calibration curve is calculated in our Statview™ Program. These are kept on file by the Quality Assurance Officer, and are also reported on the Data Sheet turned in to the Analyst.

Surrogate samples are sent to us annually by cooperating labs in the area. Analysis is performed on these samples as if they were Spikes. The Chemist does not know the concentrations of the parameters he is testing for. We expect 95% recovery of all surrogate samples. The results of our Surrogate Sampling are kept on file by the Laboratory Director, and are reported in our End of the Year Quality Control Report to the Project Director.

Many of the parameters that we test for have Alternate Methods of Analysis. Alternate Analysis is performed whenever interferences may be occurring. The Laboratory Director or the Project Director may request Alternate Testing at any time they deem necessary. For example, we routinely run three alternate methods of Ammonia analysis, the Nessler, the Phenate, and the Ion Sensitive Electrode methods.

All our reagents are made from ACS chemicals, and are stored according to specifications. They are dated and signed by the chemist who makes them. They are recorded in our Laboratory Log. They are kept on a computer inventory that is updated quarterly by the Quality Assurance Officer. All outdated reagents are discarded immediately. More importantly, since we run Equipment Blanks and Calibration Standards with every analysis, we can tell from the results of our calibration curve if our reagents are in good form.

12. Performance and System Audits

Inter-laboratory performance audits are a continuing ongoing endeavor in our research. It is of fundamental importance that our data reflects the greatest possible accuracy. Our equipment is thoroughly checked out each time it is used with a complete set of calibration standards. Calibrations are performed at least once a month on all equipment. If any instrument fails to calibrate we rely on the trained and certified staff of Florida State University technicians in our Machine and Electronic Shops to repair the equipment. Furthermore we submit to on site external equipment audits by DER on all projects.

13. Preventive Maintenance

Preventive maintenance is the key to success in any experimental endeavor. Properly performed it saves alot of time by minimizing down time of equipment. If equipment does go down it is imperative to have back-up systems which can take over.

Field equipment needs the most preventive maintenance. All meters used in the field needs to be cleaned thoroughly when they comes back to the laboratory. Field probes need to be cleaned, their membranes need to be changed, and they need to be filled with the appropriate fluids. When meters and probes go to the field all batteries need to be checked. Then the instruments are calibrated to assure that they are working properly. Two sets of meters and probes are always sent into the field so that we have backups.

Laboratory equipment are maintained as follows. Balances (two mettlers) are protected from shock, and are cleaned after daily use. All maintenance, Quarterly calibrations, are performed by certified personnel in our machine shop. Spectrophotometers (two Bausch and Lomb, two Beckman DB/G, and a Zeisse) are protected from liquid spills. They are cleaned (the optics) and calibrated with every use. If they will not calibrated they are sent to our electronics shop were certified technicians repair them. Ovens are cleaned after use and calibrated quarterly with a NBS thermometer. Water traps are used with our vacuum pumps to protect them from damage. Oil is replaced quarterly. Desiccators are kept freshly charged with drrite. Colorimeters and Turbidometers are calibrated with every use, and their optics are kept clean. All laboratory chemicals are dated, and the chemist that mixes them affixes his name to the bottle. The reagents are checked quarterly to make sure that they have not gone bad.

If any equipment goes down we have backups for them. All are kept at optimum working capacity so that they are ready to use when they are needed.

14. Specific Routine Procedures Used to Assess Data Precision, Accuracy, and Completeness.

All samples are taken in triplicate. All analysis is performed with a minimum of 10% replicates. Every time samples come to the laboratory we require field spikes, and field blanks.

The spikes and blanks assure us that the sampling was performed correctly. A good field blank which is devoid of the parameters analyzed shows that there was no contamination of the samples. Field Spikes with a Percent Recovery above 90% shows that there was no degradation of the sample before analysis. If the Percent Recovery is below 90% corrective action must be taken.

All replicates are subjected to statistical analysis. The mean is calculated for each pair of replicates are calculated as follows:

$$\text{Mean} = M = (x_1 + x_2)/2$$

The standard deviation is calculated for each set of replicates as follows:

$$\text{Standard Deviation} = S = [\sum(x_i - m)^2 / (n-1)]^{1/2}$$

The Percent Relative Standard Deviation for each set of replicates is calculated as follows:

$$\text{Percent Relative standard Deviation \% RSD} = (S / M)(100\%)$$

For any experimental analysis the mean Standard Deviation and the mean Percent Relative Standard Deviation of all the replicates for a given parameter are calculated. If it does not conform to the QA objectives of table 5 corrective action must be taken.

15. Corrective Action

The Percent Relative Standard Deviation and the Percent Recovery of Spikes are the criterion on which will be based the possibility of corrective action. If the Percent Relative Standard Deviation exceeds the values stated in section 5, or if the Percent Recovery of Spikes is less than 90% the QA Officer shall notify the Project Director who shall decide the course of corrective action. Extremely high experimental values shall also be brought to the attention of the Project Director by the QA officer.

The Project Director may decide that the data in question is worthless. If there are mistakes in the calculations the Project Director will consult with the Data Analyst (Jane Jimain), or if the experimental analysis is in question the Project Director will consult with the Chemist (Sean McGlynn).

Corrective Action may also be initiated by the DER QA Office, Performance Audits, System Audits, Laboratory/Interfield comparison Studies, or QA project Audits conducted by DER.

16. Quality Assurance Reports to Management

Quarterly Performance of Systems and Data Quality Reports will be submitted to the Project Director by the Head Chemist (Sean McGlynn). These reports include:

- A statistical assessment of Data the Percent Relative Standard Deviation and completeness of analysis.
- Results of Performance Audits
- Results of Systems Audits
- Significant QA problems and recommended solutions
- The outcome of corrective actions

Copies of all QA reports are submitted to the DER QA office.

17. Personnel Qualifications, Resumes

Curriculum Vitae: Robert J. Livingston (March, 1989)

136b Conradi
Department of Biological Science
Florida State University
Tallahassee, Florida 32306
Office: (904) 644-1466
Home: (904) 893-1453

Education

Princeton University - A.B. (1955-1959)
Columbia University - B.S. - equivalent (1961-1963)
Scripps Institution of Oceanography (University of California, San Diego)
(1963)
Institute of Marine and Atmospheric Sciences (Univ. of Miami) - M.S., Ph.D.
(1964-1970)

Fellowships and Honors

Undergraduate Fellowship - Princeton University
Graduated cum Laude, Princeton University
Dean's List - Columbia University
Aerojet General Fellowship, Scripps Institution of Oceanography (U. of
California)
Robert E. Maytag Fellowship, Institute of Marine & Atmospheric Sciences
(Univ. of Miami)
Third Annual Conservator Award; Apalachicola Seafood Festival
Conservation Award; Woodmen of the World, Life Insurance Society
Florida Scientist of the Year (1982); Museum of Science and Industry
(Tampa, Florida)

Academic and Research Positions

Research Aide (Institute of Marine & Atmospheric Sciences; 1964)
Graduate Assistant (Institute of Marine & Atmospheric Sciences; 1967-1970)
Assistant Professor (Department of Biological Science, Florida State
University; 1970-1976)
Associate Professor (Department of Biological Science, Florida State
University; 1977-81)
Professor (Department of Biological Science, Florida State University;
1981-present)
Director, Center for Aquatic Research and Resource Management (1984-
present)

Research Activities and Interests

The overall research effort of R. J. Livingston for the past 18 years has involved continuous, long-term analyses of various river and coastal systems with an emphasis on the north Florida Gulf Coast (Apalachee Bay, Apalachicola Bay). Coupled with laboratory and field experimentation, this work has included multidisciplinary systems analyses, population/community structure, trophic interactions, and the impact of various forms of anthropogenous stress on physico-chemical and biological processes. Currently, the research effort involves a comparison of 8 drainage systems (Florida, Mississippi, Alabama, Georgia, South Carolina), experimental ecology of predator/prey relationships and the validation or verification of bioassay results with field data from rivers and coastal areas. The establishment of the Center for Aquatic Research and Resource Management will provide a basis for finalization of previous studies and the initiation of new directions for the application of scientific research to resource management problems.

- Ecological (field) study of Florida Bay and inshore waters of the Florida Everglades (P.I., Dr. Durbin Tabb)
- Acute toxicity studies of effects of Dieldrin on estuarine fishes of south Florida (P.I., Dr. C. Richard Robins, Dr. Richard Wade)
- Behavioral and ecological studies on circadian rhythms of reef fishes (P.I., R. J. Livingston)
- Studies on the long-term physiological effects of chlorinated hydrocarbons and oil on estuarine organisms (P.I., R. J. Livingston)
- Laboratory studies on the effects of inorganic mercury on fresh water fishes (P.I., R. J. Livingston)
- Field and laboratory studies on the impact of pulp mill effluents on coastal organisms, Apalachee Bay (P.I., R. J. Livingston)
- Field studies on the effect of dredging and eutrophication on areas of Escambia Bay (P.I., R. J. Livingston)
- Field and laboratory studies on the impact of pesticides on the behavior and ecology of estuarine organisms, Apalachicola Bay (P.I., R. J. Livingston)
- Ecosystems management and the application of scientific data to planned development of estuarine and coastal systems (P.I., R. J. Livingston)
- Field and laboratory studies concerning the effects of clearcutting and storm water runoff on estuarine populations and communities (P.I., R. J. Livingston)
- Analysis of recovery of bay systems previously affected by anthropogenous disturbance with an emphasis on productivity, trophic structure and community response (P.I., R. J. Livingston)
- Ecosystem functions in coastal areas of north Florida (P.I., R. J. Livingston)
- Trophic relationships of coastal species associations (P.I., R. J. Livingston)
- Comparison of the effects of pulp mill wastes on drainage systems in Florida, Mississippi, Alabama, and South Carolina.
- Experimental ecology and the validation concept with respect to the impact

of toxic substances on aquatic systems. Dr. Livingston has been chosen to lead a team from the E.P.A., the Virginia Institute of Marine Science, and F.S.U. to test the validation hypothesis (P.I., R. J. Livingston)

Validation of laboratory and field experimental data for 3 river systems in the southeastern U. S. (P.I., R. J. Livingston)

Comparative analysis of long-term, multidisciplinary data in a series of river-estuarine systems.

Ecosystem analysis of the Choctawhatchee River-Bay system.

Ecological studies of Lake Jackson, Florida.

Research Grants and Funding: R. J. Livingston, Principal Investigator

F.S.U. COFRS: \$5,000 (Effects of Inorganic and Organic Mercury on Fish Physiology and Behavior; 1971-1973)

Florida Department of Transportation: \$19,800 (Effects of Dredging and Eutrophication on Escambia Bay)

National Oceanic and Atmospheric Administration (Sea Grant): \$134,000 (Estuarine Analysis: Field and Laboratory Studies on Chronic Effects of Pesticides and Other Pollutants on Estuarine Animals and Communities; 1972-1974)

Florida Department of Pollution Control: \$5,500 (Study of the Impact of a Cattle Ranch on the Apalachicola Drainage System; 1973)

Board of County Commissioners, Franklin County: \$57,000 (Physico-chemical and Biological Analysis of Apalachicola Bay; 1972-1977)

Coastal Coordinating Council, Florida Department of Natural Resources: \$5,800 (Effects of Pulp Effluent on Apalachee Bay; 1972)

Buckeye Cellulose Corporation: \$32,000 (Determination of Recovery of Heavily Polluted Portions of Apalachee Bay after Cessation of Pulp Mill Effluents; 1974-1975)

Buckeye Cellulose Corporation: \$15,000 (Study of the Impact of the Storm Water Runoff of a Clear-cut Area on the Apalachicola Drainage System; 1974-1975)

National Oceanic and Atmospheric Administration (Sea Grant): \$80,000

Franklin County Board of County Commissioners: \$24,000 (Energy Relationships and Role of Dissolved Nutrients and Detritus in the Productivity of the Apalachicola Drainage System; 1975-1976)

N.O.A.A. (Florida Sea Grant, with D.C. White) \$73,506 (Impact of Storm Water Runoff on Apalachicola Bay; 1977)

Environmental Protection Agency: \$128,000 (Analysis of Statistical Methods Used to Determine Effects of Pollutants on Aquatic Populations and Species Assemblages; 1974-1976)

National Commission on Water Quality: \$10,000 (As associate investigator with Dr. Frederick Bell, Dept. of Economics) (An Assessment of the Economic Benefits Which Will Accrue from Incremental Improvements in the Quality of Coastal Waters)

Florida Department of Environmental Regulation: \$4,000 (Study of the

Impact of Clearcutting on a Bay System; 1975)
 General Development Corporation: \$5,000 (Computerized Analysis of Estuarine Data; 1977)
 U.S. Environmental Protection Agency: \$100,000 (Analysis of Trophodynamic Processes, Apalachee Bay; 1977-78)
 Buckeye Cellulose Corporation: \$30,000 (Field and Laboratory Analysis of Factors controlling Grassbed Productivity in Apalachicola Bay; 1978)
 Florida Department of Environmental Regulation: \$10,000 (Analysis of Forestry Operations on Apalachicola Bay, 1978)
 Franklin County Board of County Commissioners: \$30,000 (Study of Impact of Clearcutting; 1977-78)
 Environmental Protection Agency: \$6,000 (Ground Truth Data Supporting Remote Sensing Analysis of Apalachicola Estuary; 1977-78)
 N.O.A.A. (Florida Sea Grant): \$81,000 (Development of Models Concerning long-term (8-year) Changes in the Biota of the Apalachicola Estuary; 1978-79)
 Thompson Hayward Chemical Company: \$24,000 (Life cycle toxicity tests; long-term effects of dimilin on *Fundulus heteroclitus*; 1977-78)
 National Science Foundation (Ecology Program, Washington, D.C.): \$36,450 (with Drs. D. White, D. Simberloff, and D. Strong) (Specialized Research Equipment Program, Instrumentation for Biochemical Analysis in Ecosystems; 1978-79)
 Franklin County Board of County Commissioners: \$20,000 (Analysis of Long-term Changes in the Physical, Chemical, and Biological Function of the Apalachicola Bay System; 1980)
 U. S. Department of State: Secretariat to the U. S. National Commission for UNESCO: \$3000 (Application of Scientific Data to Planning and Management Processes; 1980)
 U. S. National Science Foundation: \$3000. Secondary Science Training Program, Lectures (1975-80)
 Conservation Foundation (Washington, DC): \$5000 (Use of Scientific Data to Aid in Development of a Comprehensive Management Program for Franklin County, Florida; 1980)
 International Paper Company: \$720,000. (Comparison of the impact of paper mill discharges on drainage systems in Mississippi, Alabama, and South Carolina; 1980-81)
 Coastal Plains Regional Commission (Florida Department of Community Affairs): \$25,000 (Apalachicola Critical Habitat Assessment)
 N.O.A.A. (Florida Sea Grant): \$90,000 and Franklin County Board of Commissioners: \$40,000 (Management Plan for the Apalachicola Estuarine Sanctuary; 1981-82)
 Coastal Plains Commission: \$25,000, 1 year (Apalachicola Critical Habitat Assessment; 1981)
 N.O.A.A. (Florida Sea Grant and the Office of Coastal Zone Management): \$90,000, 2 years (Development of an Apalachicola Resource Atlas; 1981-82)
 Franklin County Commission: \$20,000 (Matching funds for N.O.A.A. grant; 1981-82).
 Buckeye Cellulose Corporation: \$7,000, 3 months (Review of information

concerning the Flint River in Georgia; 1982)

Florida Department of Environment Regulation: \$15,000, 15 months
(Analysis of the impact of dredging on Apalachicola Bay; 1982-83)

U. S. Environmental Protection Agency: \$1,200,000 (Field and Semi-field
Validation of Laboratory-derived Aquatic Test Systems; 1982-84) with
D. White and R. Diaz.

Franklin County Commission: \$20,000 (Studies in the Apalachicola Bay
system; 1983)

U. S. Environmental Protection Agency: \$380,000 (with W. Cooper,
Department of Chemistry) (Validation Studies in three river
systems of the southeastern U. S.; 1983-1985)

Philadelphia Academy of Natural Sciences: \$37,800: (Ecosystem stu-
dies in the Flint River system, Georgia; 1983)

U. S. Environmental Protection Agency: \$40,000 (Characterization of
offshore grassbeds in the Gulf of Mexico and as background for
offshore validation experiments; 1983-1984)

Florida Legislature through the Florida Department of Natural Resources and
the Franklin County Commission: \$69,000 (Identification and analysis
of sources of pollution in the Apalachicola Bay system; 1983)

Florida Resources and Environmental Analysis Center: \$6,800 (Water
quality/sediment analysis of Old Pass Lagoon, Destin, Florida; 1983-
1984)

Franklin County Board of Commissioners: \$40,000 (Ecology of the
Apalachicola Oyster Beds; 1984-1986)

U. S. Environmental Protection Agency: \$350,000 (with R. J. Diaz and
D. C. White) (Validation of estuarine microcosms and effects of
toxic substances on laboratory-field assemblages of
macroinvertebrates; 1984-85)

Man in the Biosphere Program, U. S. Department of State: \$4000
(Computer analysis of long-term multidisciplinary field data; 1985-86)

Florida Legislature: \$150,000 (Ecosystem analysis of the Choctawhatchee
River-Bay system; 1985-86)

Florida Department of Environmental Regulation: \$100,000 (Impact of toxic
waste sites on rivers; 1985-86)

U.S. Environmental Protection Agency: \$200,000 (with R. J. Diaz)
(Validation of estuarine microcosms; 1985-86)

Florida State University: \$150,000 (Establishment of the Center for
Aquatic Research and Resource Management; 1984-87)

U.S. Army Corps of Engineers: \$9,500 (Analysis of salinity-defined popula-
tions in the Apalachicola estuary; 1985)

Florida State University (SRAD grant): \$1,325 (Update of microcomputer
equipment)

Florida State University (summer research grant; COFRS): \$1,500 (Analysis
of oyster data)

Florida Department of Environmental Regulation: \$100,000 (Development of
a conference concerning The Rivers of Florida; 1986-87)

Florida Department of Community Affairs: \$30,000 (Biological charac-
terization of Choctawhatchee Bay, Florida; Renaissance Plaza, Inc.,
grant; 1986-87)

Florida Department of Community Affairs: \$30,000 (Continued field research in Choctawhatchee Bay, Florida; 1987-88)

Northwest Florida Water Management District: \$165,000 (Critical habitat assessment of the Choctawhatchee River system; 1987-88)

National Oceanic and Atmospheric Administration: \$9,416.00 (with Gary Ray) (Distribution of oyster larvae in Apalachicola Bay, Florida; 1986-87)

Florida Department of Environmental Regulation: \$42,000 (Chemical analysis of water and sediments of the Choctawhatchee River-Bay system; 1987)

U.S. Environmental Protection Agency: \$41,184 (Potential effects of long-term climate changes in the southeastern U.S.; 1987-88)

Florida Department of Environmental Regulation: \$5,991 (Atlas of diatoms and other algal forms from selected drainage areas in central and north Florida; 1987) with A. K. S. K. Prasad

Florida Department of Environmental Regulation: \$100,000 (Continuation funding for the center for aquatic research and resource management; 1987-88)

Northwest Florida Water Management District: \$1,900 (Ranking matrix for the water bodies of regional significance in northwest Florida; 1988)

Caribbean Marine Research Center: \$23,000 (Aquaculture support at the FSU Marine Laboratory; 1988)

Northwest Florida Water Management District: \$85,000 (Phase II--Choctawhatchee River Basin Assessment; 1988-89)

U.S. Environmental Protection Agency: \$5000 (Development of a river model, 1988)

Florida Institute of Government: \$30,000 (A simplified and rapid method for assessing the biological disturbances resulting from stormwater and marine discharges in estuaries, 1988-89)

Florida Department of Environmental Regulation: \$19,970 (An Atlas of Phytoplankton, 1988-89)

U. S. Department of State: \$16,500 (Wetlands research in the N.E. Gulf of Mexico, 1988-89)

Northwest Florida Water Management District: \$85,000 (Choctawhatchee River-Basin assessment-Phase II, 1988-89)

Caribbean Marine Research Institute: \$23,000 (Aquaculture support at the FSU Marine Laboratory, 1989)

U. S. Department of Commerce, NOAA: \$50,000 (Temperature/salinity profiles of a series of gulf estuaries, 1989)

Reviewed Publications

1. A volumetric respirometer for long-term studies of small aquatic animals. Livingston, R. J. 1968. *J. Mar. Bio. Ass. U.K.* 48: 485-497.
2. Acute and chronic effects of dieldrin on the sailfin mollie (*Poecilia latipinna*). Lane, C. E., and R. J. Livingston. 1970. *Trans. Amer. Fish. Soc.* 99(3): 389-395.
3. New design for the long-term respirometry of small aquatic animals.

- Livingston, R. J. 1970. *Copeia* (4): 756-758.
4. Circadian rhythms in the respiration of eight species of cardinal fishes. (Pisces: Apogonidae): Comparative analysis and adaptive significance. Livingston, R. J. 1971. *Marine Biology* 9(3): 253-266.
 5. Synergism and modifying effects. Livingston, R. J., et al. 1974. (Marine Technological Society, 1974) - Book Chapter (invited, in, *Marine Bioassays, Workshop Proceedings*. U.S. Environmental Protection Agency).
 6. Livingston, R. J., R. L. Iverson, R. H. Estabrook, V. E. Keys, John Taylor, Jr. Major features of the Apalachicola Bay system: Physiography, biota, and resource management. *Florida Scientist* 37(4): 245-271. 1974.
 7. Hooks, T. A., K. L. Heck, and R. J. Livingston. 1975. An inshore marine invertebrate community: Structure and habitat associations in the N.E. Gulf of Mexico. *Bulletin of Marine Science* 26: 99-109.
 8. Cripe, C. R., J. H. Cripe, and R. J. Livingston. 1975. An apparatus for the quantitative determination of activity rhythms of aquatic organisms using infra-red light emitting diodes. *J. Fish. Res. Bd. Can.* 32: 1884-1885.
 9. Livingston, R. J. 1975. Impact of pulp mill effluents on estuarine and coastal fishes (Apalachee Bay, Florida). *Marine Biology* 32: 19-48.
 10. Zimmerman, M., and R. J. Livingston. 1976. The effects of Kraft mill effluents on benthic macrophyte assemblages in a shallow bay system (Apalachee Bay, North Florida, U.S.A.). *Marine Biology* 34: 297-312.
 11. Livingston, R. J. 1976. Diurnal and seasonal fluctuations of estuarine organisms in a north Florida estuary: Sampling strategy, community structure, and species diversity. *Estuarine and Coastal Marine Science* 4: 373-400.
 12. Koenig, C. C., R. J. Livingston, and C. R. Cripe. 1976. Blue crab mortality: Interaction of temperature and DDT residues. *Archives of Environmental Contamination and Toxicology* 4(1): 119-128.
 13. Koenig, C. C., and R. J. Livingston. 1976. The embryological development of the diamond killifish (*Adinia xenica*). *Copeia*(3): 435-445.
 14. Livingston, R. J. 1976. Avoidance responses of estuarine organisms to storm water runoff and pulp mill effluents. Invited paper, Proceedings of the Third International Estuarine Research Federation conference, Galveston, Texas. October, 1975. *Estuarine Processes I.* 313-331.
 15. Livingston, R. J. 1976. Dynamics of organochlorine pesticides in estuarine systems and their effects on estuarine biota. Invited paper, Proceedings of the Third International Estuarine Research Federation Conference, Galveston

- Texas. October, 1975. *Estuarine Processes I.*, 507-522.
16. Livingston, R. J. 1976. Time as a factor in environmental sampling populations and communities. Invited paper, Symposium on the Biological Monitoring of Water Ecosystems (Ed. J. Cairns, Jr.). ASTM STP 607: 212-234.
 17. Livingston, R. J., G. Kobylinski, F. G. Lewis, and P. Sheridan. 1976. Analysis of long-term fluctuations of estuarine fish and invertebrate populations in Apalachicola Bay. *Fish. Bull.* 74(2): 311-321.
 18. Livingston, R. J., R. S. Lloyd, and M. S. Zimmerman. 1976. Determination of adequate sample size for collections of benthic macrophytes in polluted and unpolluted coastal areas. *Bull. Mar. Sci.* 26(4): 569-575.
 19. Zimmerman, M. S., and R. J. Livingston. 1976. Seasonality and physico-chemical ranges of benthic macrophytes from a north Florida estuary (Apalachee Bay). *Contr. Mar. Sci. Univ. Texas* 20: 33-45.
 20. Livingston, R. J., et al. 1977. The biota of the Apalachicola Bay system: Functional relationships. In, *Proceedings of the Conference on the Apalachicola Drainage System*. Florida Marine Research Publications, Fla. D.N.R. Eds., R. J. Livingston and E. A. Joyce, Jr., Cont. #26: 75-100.
 21. Cripe, C. R., and R. J. Livingston. 1977. Dynamics of the pesticide mirex and its photoproducts in a simulated marsh system. *Arch. Env. Cont. Tox.* 5: 295-303.
 22. Lewis, F. G., III, and R. J. Livingston. 1977. Avoidance reactions of two species of marine fishes of Kraft pulp mill effluent. *Fish. Res. Bd. Can.* 34: 568-570.
 23. Livingston, R. J. 1977. Review of current literature concerning the acute and chronic effects of pesticides on aquatic organisms. Invited paper, *Critical Reviews in Environmental Control* 7: 325-351.
 24. Livingston, R. J., et al. 1978. Long-term variation of organochlorine residues and assemblages of epibenthic organisms in a shallow north Florida (U.S.A.) estuary. *Marine Biology* 46: 355-372.
 25. Laughlin, R. A., C. R. Cripe, and R. J. Livingston. 1978. Field and laboratory avoidance reactions by blue crabs (*Callinectes sapidus*) to storm-water runoff. *Trans. Amer. Fish. Soc.* 107: 78-86.
 26. Stoner, A. W., and R. J. Livingston. 1978. Respiration, growth and food conversion efficiency of pinfish (*Lagodon rhomboides*) exposed to sublethal concentrations of bleached Kraft mill effluent. *Env. Poll.* 17: 207-218.
 27. Meeter, D. A., and R. J. Livingston. 1978. Statistical methods applied to a

- four-year multivariate study of a Florida estuarine system. Invited paper, Biological Data in Water Pollution Assessment: Quantitative and Statistical Analyses. American Society for Testing and Materials. Special technical publication 652. Eds., John Cairns, Jr., K. Dickson, and R. J. Livingston.
28. Livingston, R. J. 1978. Summary: Biological Data in Water Pollution Assessment: Quantitative and Statistical Analyses. Special technical publication 652, Editors: John Cairns, Jr., K. Dickson, R. J. Livingston. American Society for Testing and Materials.
 29. Livingston, R. J., et al. 1978. Kepone/Mirex/Hexachlorocyclopentadiene: An Environmental Assessment. Chairman, National Research Council Panel. National Academy of Sciences.
 30. Laughlin, R., R. J. Livingston, and R. Cripe. 1978. "Comments;" reply to Reynolds and Casterlin concerning laboratory/field behavior of blue crabs. Trans. Amer. Fish. Soc. 107(6): 868-871.
 31. Livingston, R. J. 1979. Multiple factor interactions and stress in coastal systems. A review of experimental approaches and field implications. In, Marine Pollution: Functional Responses. Ed. F. John Vernberg.
 32. Zimmerman, M. S., and R. J. Livingston. 1979. Dominance in benthic macrophyte assemblages from a north Florida estuary (Apalachee Bay). Bull. Mar. Sci. 29, 27-40.
 33. Livingston, R. J., and J. Duncan. 1979. Short- and long-term effects of forestry operations on water quality and epibenthic assemblages of a north Florida estuary. Ecological Processes in Coastal and Marine Systems, Ed. R. J. Livingston.
 34. Meeter, D. A., R. J. Livingston, and G. Woodsum. 1979. Short and long-term hydrological cycles of the Apalachicola drainage system with application to Gulf coastal populations. Ecological Processes in Coastal and Marine Systems. Ed. R. J. Livingston.
 35. White, D., R. J. Livingston, et al. 1979. Effects of surface composition, water column chemistry and time of exposure on the detrital microflora and associated macrofauna in Apalachicola Bay, Florida. Ecological Processes in Coastal and Marine Systems, Ed. R. J. Livingston.
 36. Sheridan, P., and R. J. Livingston. 1979. Cyclic trophic relationships of fishes in an unpolluted, river-dominated estuary in north Florida. Ecological Processes in Coastal and Marine Systems, Ed. R. J. Livingston.
 37. Livingston, R. J., and O. Loucks. 1979. Productivity, trophic interactions, and food web relationships in wetlands and associated systems. Invited paper, Proceedings of the National Symposium on Wetlands. American

Water Resources Association.

38. Stoner, A. W., and R. J. Livingston. 1980. Distributional ecology and food habits of the banded blenny, *Paraclinus fasciatus*: An inhabitant of a mobile community. *Marine Biology* 56, 239-246.
39. Livingston, R. J. 1980. Ontogenetic trophic relationships and stress in a coastal seagrass system in Florida. In, *Estuarine Perspectives*, 423-436 (Academic Press).
40. Mahoney, B. M. S., and R. J. Livingston. 1982. Seasonal fluctuations of benthic macrofauna in the Apalachicola estuary, Florida, U.S.A.: The role of predation. *Marine Biology* 69, 207-213.
41. Livingston, R. J. 1981. Man's impact on the distribution and abundance of sciaenid fishes. Sixth Annual Marine Recreational Fisheries Symposium; Sciaenids: Territorial Demersal Resources. National Marine Fisheries Service. Houston, Texas.
42. Livingston, R. J. 1981. River-derived input of detritus into the Apalachicola estuary. Proceedings of the National Symposium on Freshwater Inflow to Estuaries. U. S. Fish and Wildlife Service Publ.
43. Livingston, R. J. 1982. Trophic organization of fishes in a coastal seagrass system. *Marine Ecology, Progress Series*, 7, 1-12.
44. Livingston, R. J. 1982. Long-term variability in coastal systems: background noise and environmental stress. In, *Ecological Stress and the New York Bight: Science and Management*. U. S. Department of Commerce. pp. 605-620.
45. Dugan, P. J., and R. J. Livingston. 1982. Long-term variation in macroinvertebrate communities in Apalachee Bay, Florida. *Estuarine, Coastal and Shelf Science* 14, 391-403.
46. Greening, H. S., and R. J. Livingston. 1982. Diel variations in the structure of epibenthic macroinvertebrate communities of seagrass beds (Apalachee Bay, Florida). *Marine Ecology, Progress Series* 7, 147-156.
47. Laughlin, R. A., and R. J. Livingston. 1982. Environmental and trophic determinants of the spatial/temporal distribution of the brief squid (*Lolliguncula brevis*) in the Apalachicola estuary (North Florida, U.S.A.). *Bull. Mar. Sci.* 32, 489-497.
48. Stoner, A. W., H. S. Greening, J. D. Ryan, and R. J. Livingston. 1982. Comparison of macrobenthos collected with cores and suction dredge. *Estuaries* 6, 76-82.
49. Federle, T. W., M. A. Hullar, R. J. Livingston, D. A. Meeter, and D. C.

- White. 1982. Biochemical analysis of the spatial distribution, biomass, and community composition of microbial assemblies in estuarine, mud flat sediments. *Appl. Environ. Microbiol.* (June).
50. Clements, W. H., and R. J. Livingston. 1983. Overlap and Pollution induced variability in the feeding habits of filefish (Pisces: Balistidae) in Apalachee Bay, Florida. *Copeia* (1983), 331-338.
51. Sheridan, P. F., and R. J. Livingston. 1983. Abundance and seasonality of infauna and epifauna inhabiting a *Halodule wrightii* meadow in Apalachicola Bay, Florida. *Estuaries* 6, 407-419.
52. MacFarlane, B., and R. J. Livingston. 1983. Effects of acidified water on the locomotor behavior of the Gulf killifish. *Arch. Env. Cont. Toxicol.* 12, 163-168.
53. Federle, T. W., M. A. Hullar, R. J. Livingston, D. A. Meeter, and D. C. White. 1983. Spatial distribution of biochemical parameters indicating biomass and community composition of microbial assemblies in estuarine mud flat sediments. *Appl. Env. Micro.* 45, 58-63.
54. Livingston, R. J. 1983. Resource Atlas of the Apalachicola Estuary. Published by Florida Sea Grant College and Florida Resources and Environmental Service Center.
55. Livingston, R. J. 1983. Research and resource planning in the Apalachicola drainage system. Proceedings of the Apalachicola Oyster Industry Conference, Apalachicola. Florida Sea Grant College.
56. Federle, T. W., R. J. Livingston, D. A. Meeter, and D. C. White. 1983. Modifications of estuarine sedimentary microbiota by exclusion of epibenthic predators. *J. Exp. Mar. Biol. Ecol.* 73, 81-94.
57. Stoner, A. W., and R. J. Livingston. 1984. Ontogenetic variation in diet and feeding morphology in sympatric fishes of the family Sparidae from seagrass meadows. *Copeia*, (1984), pp. 174-187.
58. Livingston, R. J. 1984. Trophic response of fishes to habitat variability in coastal seagrass systems. *Ecology* 65, 1258-1275.
59. Livingston, R. J. 1984. Aquatic field monitoring and "meaningful" measures of stress. *Concepts in Marine Pollution Measurements.* pp. 681-691.
60. Livingston, R. J. 1985. The ecology of the Apalachicola estuary. Invited paper, U. S. Fish and Wildlife Series; Ecological Monograph.
61. Livingston, R. J. 1985. Organization of fishes in coastal seagrass systems: the response to stress. In, *Fish Community Ecology in Estuaries*

- and Coastal Lagoons. Ed., A. Yanez-Arancibia. Chapter 16:367-382.
62. Clements, W. E., and R. J. Livingston. 1984. Prey selectivity and functional response of the fringed filefish, *Monacanthus ciliatus* (Pisces: Monacanthidae). *Mar. Ecol. Prog. Ser.* 16, 291-295.
 63. Livingston, R. J., and D. A. Meeter. 1985. Correspondence of laboratory and field results: What are the criteria for verification? *Proceedings of the Symposium for Multispecies Toxicity Testing*. J. Cairns, editor. Blackburg, Virginia. Chapter 8, pp. 76-88.
 64. Livingston, R. J. 1985. The relationship of physical factors and biological response in coastal seagrass meadows. *Proceedings of a Seagrass Symposium*, Estuarine Research Foundation. *Estuaries* 7, 377-390.
 65. Livingston, R. J. 1985. Aquatic field monitoring and meaningful measures of stress. In: *Concepts in Marine Pollution Measurements*, Ed., H. H. White. pp. 681-692.
 66. Livingston, R. J., R. J. Diaz, and D. C. White. 1985. Field validation of laboratory-derived multispecies aquatic Test Ecosystems. U.S.E.P.A., Research and Development Summary Paper.
 67. Livingston, R. J. 1985. Application of scientific research to resource management: Case history, the Apalachicola Bay system. *Proceedings of the international Symposium on Utilization of Coastal Ecosystems: Rio Grande, Brazil*. pp. 103-125.
 68. Livingston, R. J. 1987. Historic trends of human impacts on seagrass meadows in Florida (invited paper). *Symposium Proceedings: Subtropical-Tropical Seagrasses of the Southeastern U.S.* Florida Marine Research Publication 42:139-151.
 69. Livingston, R. J. 1987. Field sampling in estuaries: The relationship of scale to variability. *Estuaries (Biological Variability in Estuaries)*.
 70. Livingston, R. J. 1986. Field verification of toxicity tests concerning the impact of toxic waste sites on three southeastern drainage systems. U.S.E.P.A., Research and Development Summary Paper.
 71. Federle, T. W., R. J. Livingston, L. E. Wolfe, and D. C. White. 1986. A quantitative comparison of microbial community structure of estuarine sediments for microcosms in the field. *Can. J. Microbiology* 32:319-325.
 72. Livingston, R. J. 1986 (invited paper, in press). Inshore marine habitats. In: *Ecosystems of Florida*, eds. R. L. Myers and J. J. Ewel.

73. Livingston, R. J. 1988 . Field verification of multispecies microcosms using marine macroinvertebrates. American Society for Testing and Materials. ASTM STP 971. pp. 369-383.
74. Livingston, R. J. 1987. Field verification of toxicity tests concerning the impact of toxic waste sites on three southeastern drainage systems. U.S.E.P.A. Research and Development Summary Paper.
75. Livingston, R. J. 1988. Ecological processes of recruitment in coastal epibenthic macrobiota: a comparative approach. Proceedings, IREP Workshop on Recruitment in Tropical, Coastal, Demersal Communities. Ciudad del Carmen, Mexico.
76. Livingston, R. J. 1987. Scientific research and resource management: Relationships and inconsistencies. Proceedings, National Wetland Assess. Symposium. Portland, Maine, in press.
77. Livingston, R. J. 1988. Use of freshwater macroinvertebrate microcosms in the impact evaluation of toxic wastes. Proceedings, A.S.T.M. Conference (Bal Harbour, Florida). ASTM STP 998, 166-218.
78. Livingston, R. J. 1988. Inadequacy of species-level designations for ecological studies of coastal migratory fishes. Invited paper, Sixth Biennial Conference on the Ecological and Evolutionary Ethology of Fishes. Beaumont, Texas. Environmental Biology of Fishes, 22, 225-234.
79. Livingston, R. J. 1988 Environmental research: how much is enough? Proc. Eighth Annual Minerals Management Symposium.
80. Livingston, R. J., and R. L. Howell, IV (in press). Effects of two hurricanes on oyster beds (*Crassostrea virginica*) in a gulf estuary. Estuaries
81. Ray, G. L., and R. J. Livingston (in press). The distribution of oyster larvae in Apalachicola Bay, Florida. Proc. Shellfisheries Association
82. Prasad, A. K. S. K., R. J. Livingston, and J. A. Nienow (in press). The diatom genus *Cyclotella* Kützing (Bacillariophyceae) in Choctawhatchee Bay, northeastern Gulf of Mexico: fine structure, taxonomy, and ecology.
83. Luckenback, M. W., R. J. Diaz, C. C. Koenig, R. J. Livingston, G. L. Ray, S. B. Thornton, and L. Wolfe (in review). Field validation of a multispecies benthic microcosm. I. An ecological guild approach.
84. Luckenback, M. W., R. J. Diaz, C. C. Koenig, R. J. Livingston, G. L. Ray, S. B. Thornton, and L. Wolfe (in review). Field validation of a multispecies benthic microcosm. II. Response to pentachlorophenol.
85. Ray, G. L., and R. J. Livingston (in manuscript). The short-term dynamics of estuarine polychaetes: (1) A comparison of oligohaline and polyhaline

populations.

86. Ray, G. L., and R. J. Livingston (in manuscript). The short-term dynamics of estuarine polychaetes: (2) Interannual variation in a polyhaline population.
87. Livingston, R. J. (in press, The Rivers of Florida). The Oklawaha River: Environmental Overview.
88. Livingston, R. J. and Gary L. Ray. (in press, The Rivers of Florida). Ecology of the alluvial rivers of Florida.
89. Livingston, R. J. and S. H. Wolfe. 1988. Restoration and Preservation Ranks of Water Bodies within the Northwest Florida Water Management District. Publication, Center for Aquatic Research and Resource Management
90. Livingston, R. J. 1986. Ecological processes of recruitment in coastal epibenthic macrobiota. Proc., IOC/FAO Workshop on recruitment in tropical coastal demersal communities. Report # 44, 151-166.
91. Livingston, R.J. (in press). Projected changes in estuarine conditions based on models of long-term atmospheric alteration. U. S. E. P. A. publ. CR-814608-01-0.

Other Publications

1. Living rhythms of the sea. R. J. Livingston. Sea Frontiers 14(5): 290-299. 1968.
2. Persistent pesticides and the aquatic environment. R. J. Livingston. Res. Rept. Soc. Sci. 14(2): 7-10. 1972.
3. A sea water system designed for controlled experiments on the chronic effects of pesticides on marine organisms. R. J. Livingston (with C. R. Cripe, C. C. Koenig, A. J. Tolman, and B. D. DeGrove). Sea Grant Publication, report #5, 1974.
4. Resource management and estuarine function with application to the Apalachicola drainage system (North Florida, U.S.A.). R. J. Livingston. Office of Water and Hazardous Materials, U.S. Environmental Protection Agency: included in final collection of papers (reviewed and published for submission to the Congress of the United States), Estuarine Pollution Control and Assessment, Vol. 1, 3-17. 1975.
5. Translocation of mirex from sediments and its accumulation by the hogchoker *Trinectes maculatus*. R. J. Livingston (with Gerard J. Kobylinski). Bulletin of Environmental Contamination and Toxicology 14(6): 692-698. 1975.

6. Environmental considerations and the management of barrier islands: St. George Island and the Apalachicola Bay system. R. J. Livingston. Invited paper, in *Barrier Islands and Beaches*. The Conservation Foundation. Cont. #7, 86-102. 1976.
7. Book Review: *Marine Pollutant Transfer*. R. J. Livingston. *Science* 198, 392. 1977.
8. Benthos, In: *Oil Spill Studies*. R. J. Livingston (with D. Boesch, C. Hershner, K. Roos, D. Straughan, and A. Michael (Chairman and Editor)). A.P.I. Publ. No. 4286, pp. 57-75. 1977.
9. The Apalachicola dilemma: wetlands development and management R. J. Livingston. Invited paper, National Wetland Protection Symposium; Environmental Law Institute and the Fish and Wildlife Service, U.S. Department of the Interior. 163-177. 1977.
10. Estuarine and coastal research in Apalachee Bay and Apalachicola Bay. In *Coastal Zone Management Symposium*, University of West Florida. 1977.
11. Book Review: *Effects of Pollutants on Aquatic Organisms*. R. J. Livingston. *Quart. Rev. Biol.* 1978.
12. Book Review: *Physiological Responses of Marine Biota to Pollutants*. R. J. Livingston. *Quart. Rev. Biol.* 1978.
13. Research, management, and the estuarine sanctuary concept: Where are the ties that bind? Proceedings of the workshop on the National Estuarine Sanctuary Program. The Georgia Conservancy, the Coastal Society. 1979.
14. Behavior workshop report: The role of Behavior in Marine Pollution Monitoring (with Bori L. Olla et al.). International Council of Exploration of the Sea (ICES) workshop on the problems of monitoring biological effects of pollution in the sea. *Rapp. P.-V. Reun. Cons. Inst. Explor. Mer* 179, 174-181. 1980.
15. Understanding marine ecosystems in the Gulf of Mexico. UNESCO's Man and the Biosphere Publication Series, U. S. State Department, Report No. 2, 1-8. 1980.
16. The Apalachicola Experiment: Research and management. *Oceanus* 23, 14-21. 1980.
17. Acid rain over Florida. *Geojourney* 1(2), 10-11. 1980.
18. Application of scientific data to resource management problems in coastal systems (in press). *Soviet-American Ecology Symposium (La Jolla)*.

19. Between the Idea and Reality. V.P.I. Publ. Prog. Ser. (Invited paper), 31-59.
20. Long-term biological variability and stress in coastal systems. 1982. Second U. S./U. S. S. R. Symposium; Biological Aspects of Pollutant Effects on Marine Organisms. Terskol, U. S. S. R.
21. Livingston, R. J., and S. H. Wolfe (1988). Restoration and preservation ranks of water bodies within the Northwest Florida Water Management District. Report for the Northwest Florida Water Management District.
22. Prasad, A. K. S. K. 1988. Atlas of the Microflora of North Florida Rivers. Report for the Florida Department of Environmental Regulation.
23. Livingston, R. J. The Oklawaha River System: Environmental Overview. Report for the Florida Defenders of the Environment.

Books (Editor or co-editor)

1. Synergistic Bioassays, in Marine Bioassays. Marine Technology Society, with Geraldine Cox (1974).
2. Proceedings of the Conference on the Apalachicola Drainage System. Florida Marine Research Publications (1977).
3. Biological Data in Water Pollution Assessment: Quantitative and Statistical Analyses: American Society for Testing and Materials, with John Cairns, Jr., and K. Dickson (1978).
4. Ecological Processes in Coastal and Marine Systems. Plenum Press (1979).
5. The Rivers of Florida (in press, being prepared for publication).

Professional Societies

AIBS, AAAS, American Fisheries Society, American Society of Ichthyologists and Herpetologists, American Society of Limnology and Oceanography, American Institute of Fisheries Research Biologists (invited), Gulf Estuarine Research Society, Ecological Society of America.

Papers Given at Scientific Meetings

- "Circadian Respiration Rhythms of Cardinal Fishes" (ASIH, 1970)
 Chairman, Pesticide Section (Soc. Lim. Ocean., 1972)
 "The Effects of Dredging and Eutrophication on Mulat-Mulatto Bayou (Escambia Bay, Pensacola, Florida)" (ASIH, 1973)
 "The Impact of the Pesticide Mirex on Aquatic Ecosystems" (10th Annual

- Pesticide Residue Conference, 1973)
- "The Impact of Pulp Mill Effluents on Fishes of Apalachee Bay" (Virginia Institute of Marine Science, 1973)
- "The Impact of Pulp Mill Effluents on the Aquatic Biota of Apalachee Bay, Florida" (American Fisheries Society, Annual Meeting, 1973) with Kenneth L. Heck, Theresa A. Hooks and Michael S. Zimmerman.
- "Storm Water Runoff in Estuaries" (Gulf Estuarine Research Society; Ocean Springs, Mississippi, October, 1974)
- "Resource Management and Estuarine Function with Application to the Apalachicola Drainage System (North Florida, U.S.A.)" (Invited paper, E.P.A. Conference on Estuarine Pollution Control, February, 1975, Pensacola, Florida)
- "Diurnal and Seasonal Fluctuations of Estuarine Organisms in a North Florida Estuary: Sampling Strategy, Community Structure, and Species Diversity" (ASIH, 1975)
- "The Impact of Pulp Mill Effluents on Estuarine Plant and Fish Assemblages" (Invited paper, Philadelphia Academy of Sciences; March, 1975)
- "Methods of Sampling Estuarine Systems to Determine the Long-Term Impact of Pollutants on Populations and Communities" (Invited paper, U.S. Department of Interior, Fish and Wildlife Service, Washington, D.C., April, 1975)
- "Avoidance Responses of Estuarine Organisms to Storm Water Runoff and pulp Mill Effluents" (Invited paper, the Third International Estuarine Research Federation Conference, Galveston, Texas, October, 1975)
- "Time as a Factor in Environmental Sampling Programs: Diurnal and Seasonal Fluctuations of Estuarine and Coastal Populations and Communities (Invited paper, Symposium on the Biological Monitoring of Water ecosystems, Ed. J. Cairns, Jr., et al., Blacksburg, Virginia, Nov., 1975)
- "The Impact of Pesticides on an Estuarine System" (Invited paper, Old Dominion University; Norfolk, Virginia, February, 1976)
- "Environmental Status of the Apalachicola Bay System" (Florida Defenders of the Environment, Annual meeting; Gainesville, April, 1976)
- "Impact of Organochlorine Compounds on an Estuarine System" (North Carolina State Univ. Dept. of Zoology, July, 1976)
- "Organochlorine Compounds and Long-term Changes of Estuarine Fish Populations in the Apalachicola Bay System" (Invited paper, U.S. Environmental Protection Agency officials and visiting scientist from Russia, September, 1976)
- "The Apalachicola Drainage System" (Invited paper, the Conservation Foundation, Joint Meeting of State and Federal agencies. January, 1977)
- "Applications of Scientific Data to Architectural Design in Coastal Systems" (Invited paper, School of Architecture, Florida A & M University)
- "National Stake in the Apalachicola River" (Invited paper, The Conservation Foundation, March, 1977)

- "The Apalachicola Bay System" (Invited Talk, Coastal Zone Management Advisory committee, NOAA)
- "The Apalachicola Drainage System: Functional Interrelationships" (Invited paper, Annual Meeting; Florida Defenders of the Environment, March, 1977)
- "Impact of Development on Wetlands: Case Study, the Apalachicola System" (Invited paper, National Wetland Protection Symposium, Environmental Law Institute and the Fish and Wildlife Service, U.S. Department of the Interior, June, 1977)
- "Statistical Methods Applied to a Four-Year Multivariate Study of a Florida Estuarine System" (with Duane A. Meeter) (Invited paper, American Society for Testing and Materials; John Cairns, Jr., Ed., June, 1977)
- "Temporal Variation of an Estuarine Fish Community" (American Society of Ichthyologists and Herpetologists, June, 1977), Chairman, Session on Impact of Pollutants on Fishes.
- "The Estuarine Environment" (Invited paper, Symposium on the Coastal Zone, University of West Florida; Pensacola, 17-18 June, 1977)
- "Estuarine Ecology, Impact of Forest Management" (CFM Symposium, Florida Department of Agriculture and Consumer Services; Gainesville, 17-18 August, 1977)
- "Effects of Forest Management Practices on Bay Habitat and Water Quality" (Invited paper, Apalachicola Planning and Management Program, Florida Division of Planning, Florida Division of Forestry; Bristol, 31 August, 1977)
- "Forestry Activities and the Impact on Apalachicola Bay" (Invited paper, Coastal Plains Interstate Advisory Board, Wakulla Springs, 14 September, 1977)
- "Multiple Factor Interactions: Experimental Design and Field Implications" (Invited paper, Symposium on Pollution and Physiology of Marine Organisms. Belle W. Baruch Institute for Marine Biology, South Carolina, 14 November, 1977)
- "Summary of Research Objectives" (EPA Ecological Advisory Committee, Corvallis, Oregon, 3 December, 1977).
- Plenary Address: "Coastal Ecosystems." Coastal Zone '78, Symposium on Aspects of Coastal Zone Planning and Management, San Francisco (14 March, 1978)
- Long-term Trends in the Recovery of Florida Coastal Ecosystems Affected by Pulp Mill Effluents" (Invited paper, American Fisheries Society, Annual Meeting; Kingston,)
- "Temporal Variation and Impact Analysis in Coastal Systems" (Conference on Ecological Processes in Coastal and Marine Systems, Tallahassee, Florida)
- The role of Barrier Island in the Productivity and Diversity of Lagoon Communities in Florida" (Invited paper, A.I.B.S.-Ecological Society of America, Annual Meeting; Athens, Georgia, August, 1978)
- "Long-term Changes in Coastal Systems" (Invited paper, Joint Workshop on American-Soviet Marine Research. U. S. E. P. A.; Gulf Breeze,

- Florida, September, 1978)
- "The Apalachicola Bay Ecosystem." Invited paper, Conservation Foundation (Washington, D. C.), Tallahassee, Florida (October, 1978)
- "Wetlands Food Chains." Invited paper, National Symposium on Wetlands. 14th Annual American Water Resources Conference. Lake Buena Vista, Florida (November, 1978)
- "Long-term Trends in Coastal Ecosystems." Environmental Engineering Sciences, University of Florida (March, 1979)
- "Systems Approaches to Ecological Problems." Invited paper, Australian Institute of Marine Science; Townsville, Australia (April, 1979)
- "Long-term (supra-annual) variability in coastal system--background noise and environmental stress." Invited paper, Marine Ecosystems Analysis Symposium on Ecological Effects of Environmental Stress. Estuarine Research Federation and New York Sea Grant Institute, New York (June, 1979).
- "Short-term Cycles and Long-term Trends in Two North Florida Coastal Systems." Invited paper, American-Soviet Symposium on Marine Ecol. Yalta, Russia (July, 1979)
- "Spatial/temporal Variability and Long-term Coastal Research in the N. E. Gulf of Mexico." Man and the Biosphere Program, U. S. State Department.
- "Design and Conduct of Research Programs for Marine Environment Management" (Coral Reef Workshop/Great Barrier Reef Marine Park Authority; Townsville, Australia, August, 1979)
- "Trophic Interactions of Coastal Fishes." Invited paper, International Research Conference (ERF, Biennial Meeting, October, 1979)
- "Scientific Research in Estuarine Sanctuaries." Invited paper, International Estuarine Research Conference (ERF, Biennial Meeting). Jekyll Island, Georgia (October, 1979)
- "Ecosystem Research in the N. E. Gulf of Mexico." Coastal Ecosystem Research Workshop, National Sea Grant Program; Baton Rouge, LA (November, 1979)
- "The Apalachicola River and Bay Estuarine Sanctuary." Banquet speaker, 18th Annual Southeast Regional ACM Conference; Tallahassee, Florida (March, 1980)
- "Application of Scientific Data to Resource Management Problems in Coastal Systems." Soviet-American Ecology Symposium, La Jolla (September, 1980)
- "Recent Problems in Aquatic Systems of Florida." F.L.A. Fishery Symposium, Tampa (September, 1980)
- "Hydrological Fluctuations, Detritus Movement, and Interactions of the Apalachicola Flood Plain with the Apalachicola Bay System." Panel chairman, National Symposium on Freshwater Inflow to Estuaries. U. S. Fish and Wildlife Service (September, 1980)
- "The Nature of Disturbance and Biological Variability in Estuarine Systems." Key-note Talk, Gulf Estuarine Research Society Symposium; Pensacola, Florida (October, 1980)
- "Man's Impact on the Distribution and Abundance of Sciaenid Fishes." Invited paper, Sixth Annual Marine Recreation Fisheries Symposium;

- Houston, Texas (April, 1981).
- "Development of a Regional Management Strategy for the Apalachicola Resource." Keynote talk, Regional Planning Council Annual Meeting, Blountstown (February 1981).
- "Managing Aquatic Resources: Can Science Help." Invited talk, 1-day seminar. Virginia Polytechnic Institute and State University; Blacksburg, Virginia (April, 1981).
- "The Validation Concept." Invited talk to heads of O.R.D. (E.P.A.) Laboratories (Corvallis, Athens, Duluth, Gulf Breeze, Naragansett), Gulf Breeze, Florida (May, 1981).
- "Effects of Predation on Benthic Infauna." With Bruce Mahoney. ASZ, Dallas (December, 1981).
- "Long-term Studies in Coastal Ecosystems." National Park Service Workshop on the Florida Everglades, Miami, Florida (January, 1982).
- "Seasonal Fluctuations of Benthic Infauna: Predation." With Bruce Mahoney. Benthic Ecology Meeting, Harvard (March, 1982).
- "The Application of Research to Long-term Planning and Management of the Tri-River System." Technical Workshop, Floodplain Processes in Nutrient Transport. U. S. Geological Survey (April, 1982).
- "Research Design in the Estimation of Natural Variability and Stress in Aquatic Systems." Workshop on "Meaningful Measures of Marine Pollution Effects"; N.O.A.A., E.P.A., Pensacola (April, 1982).
- "Floodplains and Wetlands--Values and Hazards of Natural Systems." Col. of Law Symposium on "Local Options for Floodplains and Wetlands Management." University of Florida, Gainesville (September 1982).
- "Apalachicola River/Bay Interactions." Apalachicola Oyster Industry Conference, Apalachicola (October, 1982).
- "Application of Research to Resource Management: Case History, the Apalachicola Estuary." Plenary Speaker, International Symposium on Utilization of Coastal Ecosystems. Rio Grande, Brazil (November, 1982).
- "Tropic Organization of a Seagrass Fish Association." Invited Paper. Oregon State University, Newport, Oregon (April, 1983).
- "Verification of Laboratory Results in the Field." Symposium on the Use of Multispecies Test Systems. Blackburg, Virginia (May, 1983).
- "Trophic Organization of Seagrass Fishes." Annual Meeting, Association of Ichthyologists and Herpetologists. Tallahassee, Florida (June, 1983).
- "The Apalachicola Experiment." Banquet Speaker (invited). Annual meeting, Association of Ichthyologists and Herpetologists. Tallahassee, Florida (June, 1983).
- "Wetland Values of the Apalachicola Drainage." Symposium on Florida's Wetlands: Florida House of Representatives. Tallahassee, Florida (August, 1983).
- "Long-term Monitoring in Coastal Systems: Research Needs and Applications." Banquet Speaker (Invited), Workshop on Monitoring Considerations in the Siting and Operation of Hazardous Waste Disposal Facilities. Tallahassee, Florida (October, 1983).
- "Coastal Seagrass Meadows: Form and Function." Invited Paper, 7th Biennial International Research Conference, Estuarine Research

- Federation. Virginia Beach, Virginia (October, 1983).
- "Field and Experimental Work in Coastal Seagrass Systems." Oregon State University Marine Science Center, Newport, Oregon (November, 1983).
- "Research and Resource Management." Corporate Weekend, Florida State University Foundation. Tallahassee, Florida (November, 1983).
- Florida's Last Natural Waterway: Can Research and Conservation Rescue It?" Invited Paper, Royal Canadian Institute. Ontario, Canada (December, 1983).
- Trophic Response and Community Structure of Macroinvertebrates and Fishes in a Coastal Seagrass System. Invited Paper, Department of Zoology, University of Texas. Austin, Texas (January, 1984).
- "Apalachicola River Basin: An Applied Study," Invited paper. 7th Annual Applied Geography conference. Tallahassee, Florida (November, 1984).
- "Effects of Barrier Island Development on Associated Lagoon Systems." Invited Paper, Conference on the Management of Developed Barriers. National Science Foundation. Virginia Beach, Virginia (January, 1985).
- "Spatial-temporal Changes in Control of Benthic Communities." Chairman, Session I on Predation/Herbivory. Benthic Ecology Meeting. Columbia, South Carolina (March, 1985).
- "Field Verification of Toxic Waste Impact on Multispecies Microcosms of Stream Macroinvertebrates." Invited paper, Ninth Symposium: Aquatic Toxicology and Environmental Fate. A.S.T.M., Philadelphia, Pennsylvania (April, 1985).
- "Ecosystem Studies in the Gulf of Mexico." Cary Arboretum, New York (April, 1985).
- "Research Perspectives in Wetland Systems." National Wetland Assessment Symposium. Portland, Maine (June, 1985).
- "Periodic Hypoxic Conditions in the Apalachicola Estuary." Invited paper. Estuarine Research Federation Conference. Durham, New Hampshire (July, 1985).
- "Scaling Factors in Estuarine Systems--A Thirteen-year Perspective." Invited paper. Estuarine Research Federation Conference. Durham, New Hampshire (July, 1985).
- "Human Impacts on Tropical and Subtropical Seagrasses of the Southeast. U.S. Botanical Society of America. Gainesville, Florida (August, 1985).
- "Analysis of Apalachicola Oyster Beds." Apalachicola River and Bay Estuarine Sanctuary Meeting. Apalachicola, Florida (October, 1985).
- "Planning and Estuarine Research." Invited participant, North Carolina Coastal and Estuarine Preplanning Meeting. Duke Marine Laboratory, Beaufort, N.C. (January, 1986).
- "Scaling and Interpretation of Laboratory Field Tests," "Field Verification of Multispecies Microcosms Using Marine Macroinvertebrates." 10th Annual Meeting, American Society for Testing and Materials. Session Chairman: "Laboratory and Field Comparisons." New Orleans (May, 1986).
- "Relationship of Laboratory Results and Field Responses of Estuarine

- Assemblages to Toxic Agents." Invited paper, Gordon Research Conference on Estuarine Processes. Plymouth, N.H. (June, 1986).
- "The Choctawhatchee River/Bay System." Invited paper, American Water Resources Association. Wakulla Spring, Florida (April, 1986)
- "Ecological Processes of Recruitment in Coastal Epibenthic Macrobiota: A Comparative Approach." Invited paper, International Conference on Recruitment in Demersal Communities. Ciudad del Carmen, Mexico (April, 1986).
- "Use of Macroinvertebrate Microcosms in the Evaluation of Toxic Waste Sites." Invited paper, ASTM meeting. Bal Harbour, Florida (November, 1986).
- "Algal Microcosms in Toxic Waste Studies: Field Verification of Laboratory Results." "Use of Research in the Management of the Apalachicola Drainage System." Invited papers, Symposium on "Algae in Freshwater Systems." Madras, India (January, 1987).
- "Inadequacy of the Use of Species-level Designations in Ecological Studies." Invited paper, Sixth Biennial Conference on Ecological and Evolutionary Ethology of Fishes. Lamar University, Beaumont, Texas (17-20 May, 1987).
- "Ecosystem Research and Resource Management." Keynote address (invited), Florida Bay Symposium. Miami, Florida (31 May-5 June, 1987).
- "Conference on the Rivers of Florida." Conference coordinator. "Apalachicola River System"; "Choctawhatchee River System" (9-10 June, 1987).
- "Ecosystem Research in the Open Ocean." Invited paper, panel member. Seventh International Ocean Disposal Symposium. Nova Scotia, Canada (21-25 September, 1987).
- "Recruiting Researchers." Invited paper, National Estuarine Research Reserve Workshop. Apalachicola, Florida (19 October, 1987).
- "Field Verification of Estuarine Macroinvertebrate Microcosms: The Use of Trophic Organization and Guilds for Predicting Toxic Impact." Invited paper. "Case Study of Application of Science to Management: The Apalachicola River-Bay System." Invited paper. "Interannual Variability of Biological Processes in Estuarine Systems: Trophic Units and Guilds." Invited paper. Ninth Biennial International Estuarine Research Conference. Estuarine Research Federation, New Orleans, Louisiana (25-27 October, 1987).
- "Impact of Climate Change on Yields of Estuarine Fisheries." Invited paper, North American Conference on Preparing for Climate Change. Washington, D.C. (27-29 October, 1987).
- "The Oklawaha: An Ecological Point of View." Invited paper, Florida Defenders of the Environment, Marion County, Florida (31 October, 1987).
- "Environmental Research: How Much is Enough?" Plenary address, invited, Eighth Annual Minerals Management Service, Gulf of Mexico OCS Region. New Orleans, Louisiana (1-3 December, 1987).
- "The Apalachicola Problem" Big Bend Sierra Club (18 April, 1988)
- "Ecological linkages: forests, rivers, and estuaries" Florida Audubon Society

- Annual Meeting. (27 October, 1988)
 "The Lake Jackson ecological situation." Council of Neighborhood Associations, Leon County. (14 November, 1988)
 "Lake Jackson ecology" Florida Wildlife Federation. (24 January, 1989)
 "Lake Jackson: Microcosm of Florida's Environmental dilemma." Florida Alumnae Association dinner. (January, 1989)
 "The ecology of Florida's estuarine systems: dark days ahead." Organization of Artificial Reefs. (9 March, 1989).

Reviewer of Manuscripts and Proposals:

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| National Institutes of Health | National Science Foundation (Population Biology and Physiological Ecology; Environmental Biology) |
| U. S. Environmental Protection Agency | |
| National Sea Grant Program (NOAA) | Virginia Journal of Science |
| National Water Quality Commission | |
| Florida Scientist Ecology | |
| Biogeography Ecological Monographs | |
| Transactions of the American Fisheries Society | Illinois Natural History Survey |
| Science | Harper & Row, Publishers, Inc. |
| Estuarine and Coastal Marine Science | Virginia Polytechnic Institute and State University (review of faculty member) |
| Northeast Gulf Science | |
| Estuaries | American Society for Testing and Materials |
| Canadian J. of Fisheries and Aquatic Sciences | Florida Department of Environmental Regulation |
| Contributions in Marine Science | |
| J. Experimental Marine Biology and Ecology | National Geographic Society |
| Marine Biology | American Fisheries Society (book) |
| Copeia | Hydrobiologia |
| Bulletin of Marine Science | American Institute of Biological Sciences (book) |
| Fishery Bulletin | Springer-Verlag |
| Archives of Environmental Contamination and Toxicology | Estuarine Research Federation |
| | Hudson River Foundation |

Teaching and Graduate Students

Courses taught at Florida State University

- Bio. 105 (General Biology), Bio. 201 (Fundamentals of Biology), Bio. 203 (Fundamentals of Ecology), Bio. 426 (Aquatic Pollution Biology), Bio. 501 (Comparative Physiology), Bio. 540 (Physiological Ecology of Fishes), Bio. 541 (Tropho-dynamics of Aquatic Systems), Bio. 646 (Advanced Ichthyology), Bio. 655 (Vertebrate Seminar), ZOO 4454C (Biology of Fishes), ISC 2937-01 (Natural Science Honors Seminar). Marine Biology

Graduate Students:

Graduated:

1. Columbus H. Brown, M.S. (The effect of photoperiodism on the respiration of the channel catfish) - presently employed as a biologist with the U. S. Fish and Wildlife Service. 1973.
2. Stephen Brice, M.S. (The effects of methyl mercury on the channel catfish *Ictalurus punctatus*) - presently employed as a research biologist with Dow Chemical Corporation. 1973.
3. Theresa Ann Hooks, M.S. (An analysis and comparison of the benthic invertebrate communities in the Fenholloway and Econfina estuaries of Apalachee Bay, Florida) - graduate of (environmental) law at F. S. U. Law School. 1973.
4. Bruce D. DeGrove, M.S. (The effects of mirex on temperature selection in the sailfin molly, *Poecilia latipinna*) - presently employed as a biologist with the Trustees of the Internal Improvement (State of Florida). 1973.
5. Kenneth L. Heck, Jr., M.S. (The impact of pulp mill effluents on species assemblages of epibenthic marine invertebrates in Apalachee Bay, Florida). 1973
6. Gerard G. Kobylinski, M.S. (Translocation of mirex from sediments and its accumulation by the hogchoker (*Trinectes maculatus*)). 1974.
7. Frank G. Lewis, M.S. (Avoidance reactions of two species of marine fishes to kraft pulp mill effluents). 1974.
8. Aureal J. Tolman, M.S. (Effects of mirex on the activity rhythms of the diamond killifish, *Adinia xenica*). 1974.
9. Claude R. Cripe, M.S. (The effects of mirex on a simulated marsh system). 1974.
10. Michael Zimmerman, M.S. (A comparison of the benthic macrophytes of polluted system (Fenholloway River) and an unpolluted system (Econfina) in Apalachee Bay, Florida). 1974.
11. Paul Muessig, M.S. (The determination of pathways of mercury concentration and conversion within specific organs of channel catfish (*Ictalurus punctatus*)). 1974.
12. Christopher C. Koenig, Ph.D. (The synergistic effects of mirex and DDT on the embryological development of the diamond killifish, *Adinia xenica*) - currently asst. professor, College of Charleston. 1975.
13. Susan Drake, M.S. (The effects of mercury on the development of the zebrafish). Fall quarter, 1975.
14. Allan W. Stoner, M.S. (Growth and food conversion efficiency of pinfish (*Lagodon rhomboides*) exposed to sublethal concentrations of bleached kraft mill effluents). Winter quarter, 1976.
15. Roger A. Laughlin, M.S. (Avoidance of blue crabs (*Callinectes sapidus*) to storm water runoff). Spring quarter, 1976.
16. George Gardner, M.S. (Behavioral reactions of pinfish to pulp mill

- effluents). Summer quarter, 1976.
17. Peter Sugarman, M.S. (Effects of bleached kraft mill effluents on activity rhythms of the pinfish, (*Lagodon rhomboides*)). Spring quarter, 1977.
 18. Bruce Purcell, M.S. (Effects of storm water runoff on grass bed communities in East Bay). Spring quarter, 1977.
 19. James Duncan, M.S. (Short-term impact of clearcutting activities on epibenthic fishes and invertebrates in the Apalachicola Bay system). Spring quarter, 1977.
 20. Peter Sheridan, Ph.D. (Trophic relationships of fishes in the Apalachicola Bay system) - presently Marine Ecologist, Bears Bluff Laboratory, U. S. EPA. Spring quarter, 1978.
 21. Allan Stoner, Ph.D. (Ecological relationships and feeding response of the pinfish, *Lagodon rhomboides*). Summer quarter, 1979.
 22. Steven Osborn, M.S. (Ecological relationships of ophiuroids in Charlotte Harbor). Fall quarter, 1979.
 23. Joseph Ryan, M.S. (Day-night feeding relationships of grassbed fishes). Spring quarter, 1981.
 24. Holly Greening, M.S. (Spatial/temporal distribution of invertebrates in grassbeds of Apalachee Bay). Summer quarter, 1980.
 25. Bruce MacFarlane, Ph.D. (Effects of variations in pH on topminnow physiology and behavior). Spring quarter, 1980.
 26. Pat Dugan, M.S. (Long-term population changes of epibenthic macroinvertebrates in Apalachee Bay, Florida). Summer Quarter, 1980.
 27. Brad McLane, M.S. (Impact of stormwater runoff on benthic macroinvertebrates). Fall Quarter, 1980.
 28. Kathy Brady, M.S. (Larval fish distribution in Apalachee Bay).
 29. Duncan Cairns, M.S. (Detrital processing in a subtropical southeastern drainage system) Fall Semester, 1981.
 30. Bruce Mahoney, Ph.D. (The role of predation in seasonal fluctuations of estuarine communities). Summer, 1982.
 31. Will Clements, M.S. (Feeding Ecology of Filefish in Apalachicola Bay, Florida). Summer, 1982.
 32. Graham Lewis, Ph.D. (Habitat Complexity in a Subtropical Seagrass Meadow). Fall, 1982.
 33. Ken Leber, Ph.D. (Feeding Ecology of Decapod Crustaceans). Summer, 1983.
 34. Kevan Main, Ph.D. (Predator-prey Interactions in Seagrass Beds: The Response of *Tozeuma carolinense*). Winter, 1983.
 35. Kelly Custer, M.S. (Gut Clearance Rates of Three Prey Species of the Blue Crab). Summer, 1985.
 36. J. Michael Kuperberg, M.S. (Response of the marine macrophyte *Thalassia testudinum* to herbivory). Fall, 1986.
 37. Susan Mattison, M.S. (The effect of post-settlement predation on community structure of epifauna associated with cockle shells). Fall, 1986.
 38. Joseph L. Luczkovich, Ph.D. (The patterns and mechanisms of selective feeding on seagrass-meadow epifauna by juvenile pinfish, *Lagodon rhomboides*). Fall, 1987.

39. Jon A. Schmidt, Ph.D. (Patterns of seagrass infaunal polychaete recruitment: influence of adults and larval settling behavior).
Fall, 1987.
40. David Bone, Ph.D. (Response of seagrass invertebrates to toxic agents). Fall, 1987.
41. Carrie Phillips, M.S. (Influence of physical disturbance on infaunal macroinvertebrates: seagrass beds vs. unvegetated areas).
Spring, 1987.
42. Frank Jordan, M.S. (Trophic organization of fishes in the Choctawhatchee River.) Spring, 1989.

Current

Jeff Holmquist (Ph.D.)
Jutta Schmidt-Gengenbach (M.S.)

Public Service

State (* = active)

Interstate 10 Environmental Study Team (1970), Florida Department of transportation.
Chairman, Select Study Committee on Mirex, Governor's Natural Resources Committee.
Advisor to Department of Transportation on Dredging and Filling Activities.
Advisor to Attorney General's Office on pollution issues.
Member, Interinstitutional Technical Advisory Committee on Environmental Affairs for Florida Pollution Control Department.
Advisor to numerous House and Senate Committees on Environmental Affairs.
Advisor to Department of Natural Resources on determination of land acquisition under the environmentally endangered lands program.
Advisor to Leon County School System for environmental education in north Florida.
Testimony before State Cabinet on environmental affairs.
Provided scientific data that went into determination of the Florida Cabinet's decision to reject a dam to be built on the Apalachicola River by the U. S. Army Corp of Engineers.
Advisor to the Calhoun County Board of Commissioners concerning environmental issues associated with the Apalachicola Bay system.
Environmental consultant on DRI for SW Florida Regional Planning Council.
Program Chairman and Speaker, Florida Defenders of the Environment Annual Meeting (1976): Controversial Waterways with Emphasis on the Apalachicola Drainage System.
Florida Defenders of the Environment Annual Meeting (1977): Three Florida Rivers.
Advisor, Florida Department of Environmental Regulation, Florida Dept. of Natural Resources, Florida Game and Fresh Water Fish Commission, Florida Division of State Planning (Department of Administration)

regarding the application of scientific data for a comprehensive management program for the Apalachicola Valley.

Reviewer (at the request of the Franklin County Board of County Commissioners) of DRI associated with development of St. George Island (1977).

Reviewer for various state agencies of reports concerning environmental impact studies (e.g., the "Chesher Report" concerning the effects of canals and development on water quality in the Florida Keys).

Trustee, Florida Defenders of the Environment. Provide scientific information for environmental problems.

Member, Citizen's Advisory Committee on Coastal Zone Management for the Apalachee Regional Planning Council.

Participant (at request of the Honorable Ralph Turlington, Commissioner of Education), Environmental Education Activities in the Apalachicola River and Bay Resource Management and Planning Program.

Scientific advisor (unpaid), Franklin County Board of County Commissioners, 1972-present.

Scientific advisor (unpaid), Wakulla County Board of Commissioners, 1980-present.

Member, Science Advisory Board, Florida League of Anglers.

Member, Shellfish Sanitation Task Force, Florida Department of Natural Resources.

Advisor (unpaid), Franklin County School Board. Development of environmental science in secondary schools.

Advisor (unpaid), Wakulla County Commercial Fisherman's Association.

Advisor (unpaid), Concerned Citizens Association of Wakulla County.

Advisor, Apalachee Regional Planning Council

Member, Board of Directors, Environmental Service Center (Florida Defenders of the Environment)

Member, Science Advisors Committee (Review of the Flint River; Chairperson: Ruth Patrick).

Member, Sanctuary Management Committee (in charge of research and education in the Apalachicola River and Bay National Estuarine Sanctuary).

Scientific advisor to Migrant Workers' Association concerning the use of the pesticide Temik in Florida

Scientific Advisor to the Florida Department of Environmental Regulation, the Audubon Society, and Getty Oil Company concerning oil drilling in the Pensacola Bay system

Scientific Advisor to the Apalachee Regional Planning Council and local citizen's groups concerning heavy metal pollution of the Chipola River

Member, Health Advisory Council, Florida Department of Health and Rehabilitative Services

*Member, Dog Island Environmental Advisory Board

Old Pass Lagoon Technical Advisory Committee (NFWMD)

Bioassay Task Force (FDER)

Member, Apalachicola Bay Area Resource Planning and Management Committee

Member, Apalachicola Bay Reserve Advisory Committee
Member, Scientific Review Committee, DOI Offshore Environmental Studies
*Member, Basin Advisory Committee, Northwest Florida Water Management District
*Member, Technical Advisory Group, Suwannee River Water Management District
*Member, Perdido Bay Cooperative Management Project.
Participant, The Florida Water Story, educational film.

Federal (* = active)

Environmental Coordinator for U. S. Environmental Protection Agency.
Coordinator, Estuarine Management Proposal, Florida Sea Grant (1973, 1974).
Advisor to U. S. Senator Lawton Chiles on environmental affairs and the energy "crisis."
Invited participant in formal presentation to the Reuss Committee on Conservation and Natural Resources (U. S. House of Representatives).
Advisor to the Select Study Committee on Mirex, Environmental Protection Agency.
Chairman, STAEP panel on Kepone/mirex/hexachlorocyclopentadiene: Reviewer of environmental and health implications for the National Academy of Sciences (Environmental Studies Board, National Research Council).
Member, Ad-hoc group to review the 1975 Water Research Strategy Doc of the Environmental Protection Agency. This included a review of research carried out by the E.P.A.
Participant in U. S. Environmental Protection Agency Conference and report (presented to the Congress of the United States) concerning Estuarine Pollution Control.
Special consultant to the National Commission on Water Quality (N.C.W.Q.) to determine the economic, social, and environmental impact of achieving or not achieving the goals of the Federal Water Pollution Control Act of 1972.
Advisor, Office of Coastal Zone Management (NOAA) concerning the eligibility of the Apalachicola Bay system for inclusion in the National Estuarine Sanctuary Program.
Coordinator (with the Conservation Foundation) of a series of meetings with federal and state agencies concerning the development of a management program for the Apalachicola Valley.
Participant in the "Operation Fish Bowl" planning and advisory group for the U. S. Army Corps of Engineers regarding the development of "Principles and Standards for Planning Water and Related Land Resources" for the Tri-River (Flint-Chattahoochee-Apalachicola) system.
Participant in the development of a joint effort with the Office of Monitoring and Technical Support (U. S. Environmental Protection Agency), the Environmental Monitoring and Support Laboratory (U. S. Environmental Protection Agency), and the National Aeronautics and

Space Administration (NASA) to apply remote sensing technology to the monitoring of stress due to upland runoff on the Apalachicola Bay system.

Member, Ecology Committee, Science Advisory Board, U. S. Environmental Protection Agency (1978-1982). (Review 5-year plan for environmental research, advise on "air/water" quality standards, etc.)

Testimony for Congressional Hearing, Barrier Islands National Parks Bill (H. R. 5981), Washington, DC (March, 1980)

Advisor, Australian Institute of Marine Science and Malcolm Fraser (Prime Minister of Australia) Research Program for the Great Barrier Reef.

Advisor, Nature Conservancy and Trust for Public Lands, Inc., concerning land purchases in Florida

Advisor to various federal environmental officers concerning environmental problems in Florida

Consultant, Environmental Effects, Transport and Fate Committee, U.S. Environmental Protection Agency

Member, Extramural Review Board, U.S. Environmental Protection Agency

Member, Habitat and Environmental Advisory Panel, Gulf of Mexico Fishery Management Council

*Member, Advisory Committee to the Man and the Biosphere Program (United Nations).

*Member, Board of Scientific Advisors, The Wetlands Fund.

Advisor, Alliance for Chesapeake Bay.

CHCOTAWHATCHEE - S. O. P.

Date 12/13/89

Page Number 1

**CHOCTAWHATCHEE DRAINAGE SYSTEM
(RIVER, BAY, OFFSHORE GULF)**

Standard Operating Procedures

(analytical protocols for water quality chemistry)

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Contents

TITLE	PAGE
I. Physical Examination.....	3
1. Temperature.....	3
2. Salinity/Conductivity.....	3
3. Dissolved Oxygen.....	3
4. pH.....	3
5. Secchi.....	4
6. Depth.....	4
II. Collection and Preservation of Samples.....	5
1. Collection	5
2. Preservation.....	6
III. Physical Characteristics.....	7
1. Apparent Color: Spectroscopic Method.....	7
2. Turbidity: Nephelometric Method.....	7
3. Total Dissolved Solids at 180°C.....	7
4. Total Suspended Solids at 103-105°C.....	7
5. Fixed and Volatile Solids at 550°C.....	8
6. Biochemical Oxygen Demand.....	9
7. Dissolved Oxygen, Azide Modified Winkler.....	10
8. Chemical Oxygen Demand.....	11
9. Particulate Organic Carbon.....	12
10. Particulate Organic Matter.....	12
11. Total Alkalinity.....	13
12. Chlorophylls, a, b, and c.....	14
IV. Nutrients.....	16
1. Nitrogen, Semi-micro Kjeldahl.....	16
2. Nitrogen, Nitrate, Cadmium Reduction.....	17
3. Nitrogen, Nitrite.....	19
4. Ammonia, Ion sensitive electrode.....	21
5. Ammonia, Phenate Method.....	22
6. Ammonia, Nesslerization.....	23
7. Phosphates, Filtration.....	25
8. Phosphates, Condensed.....	25
9. Phosphates, Total.....	26
10. Ortho-Phosphates, Ascorbic Acid.....	26
V. Solutions.....	28
1. Lugals Solution.....	28
2. Bouin's Solution.....	28
3. Probe Juice, Y.S.I., D.O.....	28
4. Probe Cleaner, Y.S.I., S.C.T.....	28
5. Probe Cleaner, Y.S.I., D.O.....	28
VI. Instruction Manuals.....	29
1. YSI Model 33 and 33M S-C-T Meters.....	29
2. YSI Model 57 Dissolved Oxygen Meter.....	30
3. Corning Calomel Electrodes.....	31

I. Physical Examination

(performed in the field)

1. Temperature

Method Number 212, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Temperature in degrees Centigrade is measured with the calibrated temperature probe in the YSI model 57 Oxygen meter. This reading is compared in the field with a similar reading from the YSI model 33 S-C-T Meter. Temperature readings are taken at the top and the bottom of the water column on site.

2. Salinity/Conductivity

Method Number 205, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Percent salinity is measured in the field with a YSI model 33 S-C-T meter calibrated in the laboratory against Standard Seawater. In waters with no appreciable salinity, conductivity in μmhos is measured with the same meter. Readings are taken at the top and bottom of the water column on site.

3. Dissolved Oxygen

Method Number 421, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Dissolved oxygen in mg/L is measured in the field with a YSI model 57 Oxygen Meter calibrated against standards analyzed by the azide modified Winkler technique. Readings are taken on site at the top and bottom of the water column. The oxygen meter is air calibrated in the field.

4. pH

Method Number 423, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

The pH is measured in pH units, the negative logarithm of the Hydrogen ion concentration, with a Corning 610A pH Meter equipped with a Calomel electrode that is calibrated in the laboratory. Readings are taken from the top and bottom of the water column in the field and in the laboratory. The meter is calibrated to pH 4, pH 7, and pH 10 with buffer solution in the field.

5. Secchi

The Secchi disk is lowered into the water slowly until the characteristic black and white pattern can no longer be distinguished. This depth is recorded in meters.

6. Depth

Depth is recorded with a graduated meter cord attached to a lead weight, which is lowered to the bottom. At contact, depth is recorded in meters.

II. Sample Collection and Preservation of Samples

1. Collection

*The result of any test can be no better than
the samples on which it is performed. ""*

An old Axiom

The objective of sampling is to collect a portion of material small enough in volume to be transported conveniently and handled in the laboratory while still accurately representing the material being sampled. This implies that the relative proportions or concentrations of all pertinent components will be the same in the samples as in the material being sampled and that the sample will be handled in such a way that no significant changes in composition will occur before the tests are made.

Sample bottles must be rinsed with the water being sampled at least three times. Sample containers that are to be re-used are rinsed at least three times with dl water, dried, and then sealed to avoid any contamination. If phosphates are to be analyzed, the use of detergents must be avoided, unless the sample containers are acid washed with warm 10% HCl.

Samples collected at a particular time and place can represent only the composition of the source at that time and place. Grab samples are collected with a Kemmerer sampler at the bottom of the water column. Avoid collecting detritus by taking the sample a few centimeters above the soil/water interface. Surface samples are collected by lowering an inverted sample container beneath the water/air interface and righting it. Avoid collecting any flotsam and jetsam by filling the sample container 5cm beneath the surface. Avoid entrapping air in the filled sample container. The sample must be kept on ice in the dark until it is received at the laboratory.

When a source is known to vary with time, samples must be taken with appropriate frequency to monitor the extent of these variations. In such a situation, the location and the time of sample collection must be accurately duplicated. In open water, a Loran can assure site location to within a hundred feet, otherwise landmarks must be judiciously chosen.

Samples are put on ice in the dark immediately to assure stability of constituents until they can be analyzed in the lab. Before delivery of the sample to the lab, a chain of custody form must be filled out detailing the volume of the sample, the location of the site, the date and time of sampling, the name of the samplers, the project and/or the parameters to be analyzed, the technique by which the sample was obtained, and the methods of preservation.

Samples are to be delivered to the lab with all possible haste, if delivery time exceeds 24 hours, correct preservation techniques must be observed. Generally, a sample will be accepted by the laboratory if they are on ice, with a proper chain of custody form. Once received samples are allowed to rise to ambient temperature before analysis.

2. Preservation

The unequivocal preservation of samples is fundamentally impossible. Regardless of the preservation technique, complete stability for every constituent can never be achieved. It is best to analyze samples as soon as possible after collection, and then to judiciously determine the type of preservation to be utilized.

Measurement	Container	Preservative	Holding
Color	P, G	Cool, 4°C	48 hours
Conductance	P, G	Cool, 4°C	28 days
pH	P, G	None	None
Filterable Residue	P, G	Cool, 4°C	7 days
Non-filterable Residue	P, G	Cool, 4°C	7 days
Temperature	P, G	None	None
Turbidity	P, G	Cool, 4°C	48 hours
Alkalinity	P, G	Cool, 4°C	14 days
Ammonia	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Kjeldahl Nitrogen	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Nitrate	P, G	Cool, 4°C	48 hours
Nitrite	P, G	cool, 4°C	48 hours
Oxygen, dissolved	P, G	None	None
Ortho-Phosphate	G	Cool, 4°C, no acid	48 hours
Total Phosphate	G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Phosphate, T. dissolved	G	Cool, 4°C, H ₂ SO ₄ to pH < 2	24 hours
BOD	P, G	Cool, 4°C	48 hours
COD	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days
Organic Carbon	P, G	Cool, 4°C, H ₂ SO ₄ to pH < 2	28 days

III. Physical Characteristics

(analyzed in the our Chemistry Laboratory)

1. Apparent Color: Spectroscopic Method

E. P. A. approved HACH™ method, and Method Number 204 B., *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Samples are allowed to settle. Decant 25ml from the sample container into the HACH™ graduated boro-silicate colorimetric vial. The color is measured against a dl water blank on one of two different colorimeters (HACH™ DR/1 or HACH™ DR/A) using an alpha-platinum-cobalt filter. Colorimetric values range from zero to 500 units. One color unit is equal to 1mg/L platinum as the chloroplatinate ion.

2. Turbidity: Nephelometric Method

Method Number 214 A., *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Fill the sample tube with sample and standards which have been homogenized, allowing all air bubbles to escape. The standard blank is turbidity-free water from HACH™ sealed in ampules measuring 0.61, 10.0, 100.0, and 1000.0 NTU. A HACH™ model 2100A turbidometer equipped with a tungsten-filament lamp and photoelectric cells to detect light scattered at 90° to the path of incident light is used. Sample vials are kept scrupulously clean and scratchless. Between samples, wash the vials with three volumes of dl water, and zero the instrument. Be sure to dry the surfaces of the vial, particularly it's base, before placing the sample in the turbidometer. Turbidity values range from 0 NTU to 1000 NTU, and roughly approximate the old Jackson Candle Turbidity units.

3. Total Dissolved Solids at 180°C

Method Number 209 B., *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

(1) Glass Fiber Filter Disk : Whatman® 4.25cm dia. 934-AH. Rough side down.

(2) Evaporating Dish : ignite at $550 \pm 50^\circ\text{C}$ for one hour in the muffle furnace to rid it of possible contaminants.

(3) 2.5-200.0mg of sample should be obtained in less than 10 minutes filtering time. More sample may lead to the entrapment of water in a thick residue.

(4) Filtration : Filter a measured volume of well-mixed sample and wash with three 10ml volumes of dl water. Transfer the filtrate to a weighed evaporation dish and heat in an oven at $180 \pm 2^\circ\text{C}$ and cool in a dessicator. Repeat until a constant weight is obtained, or until weight loss is less than 4% of the previous weight, or 0.5mg, whichever is less.

Calculations

$$\text{mg total dissolved solids/L} = \frac{(A-B) \times 1000}{\text{sample volume in ml}}$$

4. Total Suspended Solids at 103-105°C

Method Number 209 C., *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

(1) Glass Fiber Filter Disk : Whatman® 4.25cm dia. 934-AH. Rough side down. Ignite at 550°C for 15 minutes to rid the filter of contaminants. Store in a dessicator, and weigh before use.

(2) Sample: select suitable volume of sample to obtain between 2.5-200mg of sample in less than 10 minutes filtering time.

(3) Filtration: assemble filtering apparatus and begin suction. Wet filter, and filter a well-mixed volume of sample. Wash with three 10ml volumes of distilled water. Remove and dry filter in an oven for at least 1 hour at 103-105°C. Cool the filter in a dessicator. Repeat cycle until weight is constant, or weight loss is less than 4% of previous weight, or weight loss is less than 0.5mg, whichever is less.

Calculations

$$\text{mg total suspended solids/L} = \frac{(A - B) \times 1000}{\text{sample vol. (ml)}}$$

where : A = weight of filter + dried residue (mg), and
B = weight of filter (mg).

5. Fixed and Volatile solids at 550°C

Method Number 209 D., *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

(1) Sample: Oven dried residue from the Total Dissolved Solids and the Total Suspended Solids tests. These two residues must be worked up separately, thus there will be two different fixed and volatile determinations.

(2) Muffle Furnace: Ignite the sample at 550 ± 50°C for at least an hour in a muffle furnace. Cool in dessicator and repeat cycle if weight loss is non-existent, less than 4% of previous weight, or less than 0.5mg.

Calculations	
mg volatile solids/L	= $\frac{(A - B) \times 1000}{\text{sample volume (ml)}}$
mg fixed solids/L	= $\frac{(B - C) \times 1000}{\text{sample volume (ml)}}$
A = weight of dried residue + dish + filter before ignition (mg)	
B = weight of dried residue + dish + filter after ignition (mg)	
C = weight of dish + filter (mg)	

6. Biochemical Oxygen Demand

Method Number 507, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

This empirical bioassay measures the dissolved oxygen consumed by microbial life assimilating and oxidizing the organic matter present. 300, 275, 200, and 125ml of the sample are incubated for five days in the dark, at 20°C, in standard BOD bottles. The remaining portion of the BOD bottle is filled with a HACH™ BOD nutrient solution. The reduction in dissolved oxygen concentration during the incubation period yields a measure of the biochemical oxygen demand. Reagent, nutrient, dl water and glucose/glutamic acid blanks are run simultaneously, to make sure that there is no contamination from other sources. Determination of dissolved oxygen can be accomplished with either the azide modified Winkler technique, or with a YSI dissolved oxygen meter and BOD bottle probe equipped with a stirrer boot.

Run checks assuring that contamination is not affecting the BOD. The glucose-glutamic acid standard check is one of the best. Dry reagent-grade glucose and reagent-grade glutamic acid at 103°C for one hour. Take 150 mg of each and dilute to one liter. This solution must be prepared fresh immediately before use. If the 5d 20°C BOD value of the glucose-glutamic acid standard check (use a 2% dilution) is less than 200 ± 37mg/L, reject any BOD determinations. Run a nutrient blank and a dl water blank as additional checks.

Calculations	
BOD, mg/L	= $\frac{D_1 - D_2}{P}$
D ₁ = DO of diluted sample immediately after preparation, mg/L.	
D ₂ = DO of diluted sample after 5d incubation at 20°C, mg/L.	
P = decimal volumetric fraction of sample used.	

Standard Deviation = 0.120mg/L

7. Dissolved Oxygen: Azide Modified Winkler

Method Number 421 B, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Manganous Sulfate Solution: 364g $MnSO_4 \cdot H_2O$ in distilled water, filter and dilute to 1L.
- (2) Alkali-iodide-azide reagent: 500g NaOH (or 700g KOH) and 135g NaI (or 150g KI) in distilled water, dilute to 1L. Add 10g NaN_3 dissolved in 40ml distilled water. Should not give color with starch solution when diluted or acidified.
- (3) Sulfuric Acid: concentrated.
- (4) Starch: 2g laboratory-grade soluble starch and 0.2g salicylic acid in 100ml distilled water.
- (5) Standard Sodium Thiosulfate titrant: 6.205g $Na_2S_2O_3 \cdot 5H_2O$ in distilled water. Add 1.5ml 6N NaOH or 0.4g of solid NaOH and dilute to 1000ml. Standardize with bi-iodate solution.
- (6) Standard Potassium bi-iodate solution: 812.4mg $KH(IO_3)_2$ in 1000ml distilled water. To standardize, dissolve 2g KI in 100-150ml water. Add 1ml 6N H_2SO_4 and 20ml of standard bi-iodate solution. Dilute to 200ml, and titrate liberated iodine with thiosulfate titrate to a pale straw color. Add starch, and continue titrating until blue color disappears.
- (7) Potassium Fluoride Solution: 24.689g of anhydrous KF in 100ml H_2O .

Procedure

- (1) To sample add 1ml $MnSO_4$ solution.
- (2) Add 1ml alkali-iodide-azide reagent.
- (3) Stopper to exclude air bubbles and mix by inverting bottle.
- (4) After ppt settles to about 1/2 volume of bottle, leaving a clear supernate, add 1ml conc. H_2SO_4 . Restopper and mix.

Calculations

- (1) For titration of a 200ml sample, 1ml 0.0021 M $Na_2S_2O_3 = 1mg DO/L$
- (2) To express results in percent saturation at 1 atm pressure (101.3 kPa.), use solubility data in the table on the next page. Warning: Brown-colored waters containing tannic acid and humic acids interfere with the Winkler test; on such waters a D.O. meter must be used.

Standard Deviation = $20\mu g/L$

8. Chemical Oxygen Demand, Closed Reflux, Colorimetric

Method Number 508 C, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents^o

- (1) Digestion Solution : 10.216g $K_2Cr_2O_7$ (dried at 103C for 2 hours) in 500ml dl water. Add 167ml concentrated H_2SO_4 slowly with stirring, and then add 33.3g $HgSO_4$. Let all the components dissolve, cool to room temperature, and then dilute to 1000ml.
- (2) Silver/Sulfuric Acid Solution : Add Ag_2SO_4 crystals or powder to concentrated H_2SO_4 , at a rate of 5.5g Ag_2SO_4/kg H_2SO_4 (5.5g/544.8ml). Let it stand for 1 to 2 days to dissolve the Ag_2SO_4 .
- (3) Potassium Hydrogen Phthalate (KHP) standard : Lightly crush and dry the KHP to constant weight at 120°C Dissolve 425mg in dl water and dilute to 1000ml. Stable when refrigerated for up to 3 month in the absence of any visible biological growth.

Procedure

- (1) Homogenize sample at high speed for 2 minutes. This insures a uniform distribution of suspended solids, and thus improves the accuracy and reproducibility of test results.
 - (2) Prepare reaction vials in the optical grade screw-cap vials.
Silver/Sulfuric Acid Reagent..... 3.5ml
Digestion Solution..... 1.5ml
Sample Size..... 2.5ml
 - (3) Holding the vial by the cap in an empty sink, swirl the vial using a circular wrist motion, until the contents are mixed.
 - (4) Place the vial in the preheated COD reactor. Reflux for 2 hours at 150°C.
 - (5) Allow the vials to cool then measure their absorbance in the HACH™ DR/1 Colorimeter.
- Standard Deviation = 3mg/L

III. Physical Characteristics, cont.

9. Particulate Organic Carbon

Method 3.1, *A Manual of Chemical and Biological Methods for Seawater Analysis*, Parsons, Maita, and Lalli. 1984.

Reagents

- (1) Sulfuric Acid Dichromate : 4.84g of $K_2Cr_2O_7$ in 20ml of dl water. Add this solution, a little at a time, to approximately 500ml of concentrated sulfuric acid. Cool to room temperature and bring the total volume up to one liter with more concentrated sulfuric acid in a volumetric flask. Warning... this reagent is particularly caustic.
- (2) Phosphoric Acid : Analytical grade 70% phosphoric acid.
- (3) Sodium Sulfate Solution : Dissolve 45g of anhydrous Na_2SO_4 in 1000ml of dl water.
- (4) Stock Glucose Solution : Dissolve 7.50g of pure glucose, and a few crystals of $HgCl_2$, in distilled water and bring up to a final volume of 100ml. The solution is stable for many months in a refrigerator, but should be discarded if any turbidity results.
- (5) Standard Glucose Solution : Dilute 10.0ml of Stock Glucose Solution to 1L in dl water, 1.00ml=100 μ g of carbon.

Procedures

- (1) Place a Whatman® 4.25cm dia. 934-AH glass microfibre filter in the Millipore® filter apparatus. Attach a controlled vacuum source not exceeding 1/3 atmosphere. After filtration of a suitable volume of sample, usually 0.5L to 2.0L, apply full suction to the filter. Release the suction after 1 minute, add 2ml of the sodium sulfate reagent, and reapply the suction immediately ; repeat this process once more with 2ml of sodium sulfate and remove the filter under suction.
- (2) Place the filter into the bottom of a 50ml beaker. Add 1.0ml of phosphoric acid and 1.0ml of distilled water. Mix and place into a block heater at 100-110°C for 30 minutes. Cover with a watch glass during this period.
- (3) Add 10ml sulfuric acid-dichromate oxidant and 4ml of dl water.
- (4) Mix by swirling and place replace watch glass cover. Heat for 60 minutes at 100-110°C.
- (5) Allow the mixture to cool. Transfer the solution and the filter pad to a 50ml graduated cylinder. Rinse the sides of the beaker beaker with dl water. Add this wash to the cylinder. Stopper and mix by inverting. ; allow solution to cool, and the filter should settle on the bottom.
- (6) Measure the extinction of the blank against the sample at 440nm. As the blank will have a higher absorbance than the sample, the normal placement of samples in the spectrophotometer will need to be reversed. Zero the sample against air.

Calculations

To determine the particulate organic carbons by this "wet ashing" technique there must be a comparison of the decrease in the extinction of the sample solutions with known standards. Prepare these standards, bringing them up to a volume of 50ml with

dl water, use them to calibrate every run of samples, and plot a concentrations versus absorbance curve. Calculate sample concentrations from the slope of the standard line.

Standard Number	ml standard glucose	mg C
1	0	0
2	1.0	0.30
3	2.0	0.60
4	3.0	0.90
5	4.0	1.20
6	5.0	1.5
7	10.0	3.00

10. Particulate Organic Matter

This is the same as the Total Suspended Volatile Solids. Method Number 209 D, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

11. Total Alkalinity: titration to an endpoint of pH4.5

Method Number 310.1, *Methods for Chemical Analysis of Water and Wastes*, EPA, 1983.

Procedure

Sample should be stored at 4°C and run as soon as possible. Do not open the sample bottle before analysis. Use a large volume of titrant for greatest accuracy, but keep volume low enough to assure a sharp endpoint. Place sample in flask by pipetting with the pipet tip near the bottom of the flask. Add standard acid while stirring with a Teflon coated magnetic stirrer. Allow the pH meter to obtain equilibrium. Keep the air space above sample to a minimum, the electrode and buret in contact with the sample may be covered with parafilm or inserted through a rubber stopper. Titrate to a pH of 4.5. Record the volume of titrant.

Reagents

(1) Sodium Carbonate Solution : 0.05N, 2.5 + 0.2g Na₂CO₃ dissolved in dl water in a 1L volumetric flask. The sodium carbonate must be dried at 250°C for 4 hours and cooled in a dessicator prior to weighing.

(2) Standard Acid : 0.1N, HCl, or H₂SO₄. Dilute 3.0ml conc. H₂SO₄ or 8.3ml conc. HCl to 1L with dl water. Standardize with 40ml of 0.05N Na₂CO₃ solution with about 60ml of dl water by titrating potentiometrically to pH 5. Lift the electrode and rinse it off into the beaker. Boil the solution gently for 3-5 minutes under a watch glass cover. Cool to room temperature. Rinse the cover off into the solution. Continue the titration to the endpoint.

Calculations

$$\text{Normality, N} = \frac{A \times B}{53.00 \times C}$$

A = grams Na₂CO₃ weighed into 1L

B = ml Na₂CO₃ solution

C = ml acid used to reach endpoint

$$\text{Alkalinity, mg/L CaCO}_3 = \frac{D \times N \times 50.000}{Z}$$

D = ml of standard acid

N = normality

Z = ml of sample

Standard Deviation = 3.0mg/L Calcium Carbonate.

12. Chlorophyll

Method 4.1, *A Manual of Chemical and Biological Methods for Seawater Analysis*. T. Parsons, Y. Maita, C. Lalli, Pergamon Press, 1984.

A known volume of seawater is filtered onto a synthetic filter or onto a glass fiber filter; pigments are extracted from the filter in 90% acetone and their concentration is estimated spectrophotometrically.

Between 0.5 and 1 liter of seawater are filtered through a membrane or glass fiber filter (pore size .45 μ). As the seawater is being filtered, a few drops of a suspension of magnesium carbonate in seawater are added to prevent acidity on the filter.

The filter is drawn dry, removed, and can be folded and stored in a desiccator at -20°C for a least 30 days if analysis can not proceed immediately. Filters should be folded in half, backed with a piece of ordinary paper and fastened with a paper clip for storage.

Reagents

(1) 90% acetone: With a graduated cylinder measure 100ml of dl water into a 1000ml volumetric flask. Bring the volume of liquid in the flask to 1000ml with analytical grade acetone. The reagent needs to be stored in a tightly stoppered bottle in the dark.

(2) Magnesium Carbonate: Add 1g of powdered MgCO₃ to 100ml of dl water, and shake vigorously.

Procedure

(1) Invert a polyethylene bottle containing the seawater sample into the Millipore filtering equipment containing a membrane or fiber glass filter. Allow the sample to filter under 1/2 atmosphere pressure vacuum.

(2) Add several (3 to 5) drops of MgCO₃ solution to the seawater as it is being filtered.

(3) Drain the filter thoroughly with the suction and store or extract as necessary.

CHOCTAWHATCHEE - S. O. P.

Date 12/13/89

Page Number 15

- (4) Place the filter in a 15-ml centrifuge tube; add 15ml of 90% acetone to volume and shake thoroughly. Allow to stand overnight in a dark place (preferable refrigerated).
 (5) Centrifuge the contents of each tube at room temperature for 5 to 10 min--the exact time depending on the model of centrifuge and the degree of clarity obtained (optical density at 750 nm should be less than 0.05 in a 10-cm cuvette).
 (6) Decant the supernate into a 10-cm path length spectrophotometer cuvette and measure the extinction at the following wavelengths without delay (sample should be at room temperature to avoid misting on the optical cell).

Wavelengths: 750, 664, 647, and 630.

- (7) Correct each extinction for a small turbidity blank by subtracting the 750 nm from the 664, 647, and 630 nm absorptions. (The 510 nm absorbance is corrected by subtracting 2X and 750 nm absorbance.)

Calculations

Calculate the amount of pigment in the original seawater sample using the equations given below:

$C_a =$	Chlorophyll a = 11.85 E_{664} - 1.54 E_{647} - 0.08 E_{630}
$C_b =$	Chlorophyll b = 21.03 E_{647} - 5.43 E_{664} - 2.66 E_{630}
$C_c =$	Chlorophyll c = 24.52 E_{630} - 1.67 E_{664} - 7.60 E_{647}

where E stands for the absorbance at different wavelengths obtained above (corrected by the 750 nm reading) and C_a , C_b and C_c are the amounts of chlorophyll (in $\mu\text{g/ml}$ if a 1-cm light path cuvette is used); then:

$\frac{\text{mg chlorophyll}}{\text{m}^3} = \frac{C \times v}{V \times 10}$

where v is the volume of acetone in ml (15 ml), V is the volume of seawater in liters and C_a , C_b , and C_c are the three chlorophylls which are substituted for C in the above equation, respectively (Note $\mu\text{g/l} = \text{mg/m}^3$).

IV. Nutrients

(analyzed in the our Chemistry Laboratory)

1. Nitrogen, semi-micro KjeldahlMethod Number 420 B, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.Reagents

- (1) Absorbant solution, plain boric acid: dissolve 20g H_3BO_3 in dl water and dilute to 1L.
- (2) Borate buffer solution: 88ml 0.1N NaOH (or 4.0g NaOH in 88ml dl water to about 500ml of 0.025M sodium tetraborate (9.5g $N_2B_4O_7 \cdot 10H_2O/L$) and dilute to 1L.
- (3) Digestion reagent: 134g K_2SO_4 in 650ml dl water and 200ml conc. H_2SO_4 . Add, while stirring, 25ml mercuric sulfate solution. Dilute the combined solution to 1L with water. Keep at a temperature close to 20 degrees C. to prevent crystallization.
- (4) Sodium hydroxide - sodium thiosulfate reagent: 500g NaOH and 25g $Na_2S_2O_3 \cdot 5H_2O$ in water and dilute to 1L.
- (5) All reagents for the determination of Ammonia by Nesslerization.

Procedure

Determine desired sample size based on the following table:

Organic Nitrogen in sample mg/L	Sample Size mL
4-40	50
8-80	25
20-200	10
40-400	5

- (1) Transfer 50ml of sample, or an appropriate volume diluted to 50ml with ammonia-free water to a 125ml erlenmeyer flask
- (2) Add 3ml borate buffer solution and adjust pH to 9.5 with 6N NaOH using a pH meter. Quantitatively transfer sample to a 100ml Kjeldahl flask and boil off 30ml. This removes the ammonia. Skip this step if removal of ammonia is not desired.
- (3) Add 10ml of digestion reagent to the sample in the Kjeldahl flask. Add 5 or 6 glass beads (3-4mm diameter). Set heat on the Kjeldahl digestion rack on medium, boil briskly under hood until solution clears (a pale straw color is acceptable) and copious fumes are observed, usually about 30 minutes. Now turn heat up to the maximum setting and digest for an additional 30 minutes. Cool.

CHOCTAWHATCHEE - S. O. P.

Date 12/13/89

Page Number 17

- (4) Quantitatively transfer digested sample by diluting and rinsing into a rapid distillation unit so that the total volume does not exceed 30ml. Add 10ml of hydroxide-thiosulfate reagent.
- (5) Adjust rate of steam in rapid distillation unit so that there is no escape of steam from the tip of the condenser or bubbling of contents of the receiving flask. Distill and collect 30-40ml distillate below surface of 10ml boric acid solution contained in a 125ml distillate erlenmeyer flask. Extend tip of condenser well below level of the boric acid solution and do not let temperature in condenser rise above 29°C. Lower collected distillate free of contact with delivery tube and continue distillation during the last 1 or 2 minutes to cleanse condenser.
- (6) Carry a blank through the procedure.
- (7) Determine ammonia by nesslerization.

Standards for Kjeldahl Nitrogen (do not go through step 2)

Standard No.	ml Standard Ammonia	Conc. (mg/l)
1	0	0
2	0.5	0.122
3	1.0	0.244
4	2.0	0.488
5	4.0	1.220
6	5.0	2.440

2. Nitrogen, Nitrate, Cadmium Reduction

Method Number 418 C, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Nitrate-free water: blank not to exceed 0.01 absorbance units.
- (2) Copper-cadmium granules: Wash 25g of 40-60 mesh Cd granules with 6N HCl and rinse with water. Swirl Cd with 100ml of 2% CuSO₄ solution for 5 min. or until blue color partially fades. Decant and repeat with fresh CuSO₄ until a brown colloidal precipitate develops. Wash Cu-Cd granules 10 times with water to remove all precipitated Cu.
- (3) Sulfanilimide reagent: dissolve 5g of sulfanilimide in a mixture of 50 ml conc. HCl and 300ml dl water. Dilute to 500ml with dl water.
- (4) N-(1-naphthyl)-ethylenediamine dihydrochloride solution: 500mg NED dihydrochloride in 500ml water. Store in a dark bottle, replace monthly and make a new calibration with each batch.
- (5) Ammonium chloride-disodium ethylene diamine tetra-acetate solution: 26g NH₄Cl and 3.4g EDTA in 2L of dl water. Add 1.3L of dl water and approximately 4.5ml NH₄OH to bring to pH 8.5.
- (6) Hydrochloric acid: 6N HCl
- (7) Copper sulfate, 2%: 20g CuSO₄ 5 H₂O in 500ml H₂O, then dilute to 1L.

CHOCTAWHATCHEE - S. O. P.

Date 12/13/89

Page Number 18

(8) Stock nitrate solution: Dry KNO_3 in an oven at 105°C for 24 hours. Dissolve 0.7218g and dilute to 1 liter. Preserve with 2ml CHCl_3/L , 1ml = 100.0 $\mu\text{g NO}_3\text{-N}$. This solution is stable for at least six months.

(9) Standard nitrate solution:: Dilute 50.0ml stock nitrate solution to 500 ml with water. 1.00ml = 10.0 $\mu\text{g NO}_3\text{-N}$.

(10) Stock Nitrite Solution: 0.6072g KNO_2 (dried in a dessicator for 24 hours) is dissolved in nitrite-free water and diluted to 1L. 1.00ml = 100 $\mu\text{g NO}_2\text{-N}$. Preserve with 2ml CHCl_3 and refrigerate. It is stable for 3 months.

(11) Standard Nitrite Solution: 50.0ml stock diluted to 500ml with nitrite-free water. 1.00ml = 10.0 $\mu\text{g NO}_2\text{-N}$.

Procedure

(1) Preparation of reduction column:

Insert glass wool plug and fill column with water. Add CuCd granules to a height of at least 18.5cm. Keep water over column to prevent entrapment of air. Wash column with 200ml of dilute $\text{NH}_4\text{Cl-EDTA}$. Activate column by passing through it, at 7 to 10mls per minute, 100ml of a solution of 25ml of a 1.0mg $\text{NO}_3\text{-N/L}$ standard and 75ml $\text{NH}_4\text{Cl-EDTA}$ solution.

(2) Turbidity removal: if necessary filter through either a $0.45\mu\text{m}$ membrane or glass fiber filter.

(3) pH adjustment: adjust pH to 7-9 with dilute HCl or NaOH .

(4) Sample reduction: to 25.0ml of sample or a portion diluted to 25.0 ml, add 75ml $\text{NH}_4\text{Cl-EDTA}$ solution and mix. Pour into column and collect at a rate of 7-10ml/min. Discard first 25ml. Collect the rest in the original sample flask. There is no need to wash column between samples, but if column is not to be used for several hours or longer, pour in 50ml of dilute $\text{NH}_4\text{Cl-EDTA}$ solution, letting it pass through system. Store column in the solution; never let it dry out.

(5) Color development: As soon as possible and not more than 15 minutes after reduction, add 2.0ml sulfanilimide reagent to 50ml sample. Let react for 2-8 min. Add 2ml $\text{NED-dihydrochloride}$ solution and mix immediately. Measure absorbance at 543nm (after 10 min. but before 2 hours) against a distilled water-reagent blank.

(6) Standards: Dilute the following volumes of standard nitrate solution to 25ml with dl water, and add 75ml of dilute Ammonium-Chloride-EDTA solution. Compare at least one $\text{NO}_2\text{-}$ standard to the $\text{NO}_3\text{-}$ to verify column efficiency.

Standards for Nitrate		
Standard No.	ml Standard Nitrate	Conc. (mg/l)
1	0	0
2	0.2	0.08
3	0.5	0.20
4	1.0	0.40
5	2.0	0.80
6	5.0	2.00
7	10.0	4.00

Calculations

Compute sample concentration directly from the standard curve, report as mg oxidized N (NO_3^- plus NO_2^-) per liter. Subtract nitrite results to give figures for nitrate.

Application Range = 0.01 to 1.0 mg/L

3. Nitrogen, Nitrite Analysis

Method Number 419, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Nitrite-free water: Add a crystal of KMnO_4 to 1L distilled water. Discard initial 50mls. Collect the fraction that is free of permanganate, which gives a red color with DPD reagent.
- (2) Sulfanilimide reagent: Dissolve 5g of sulfanilimide in a mixture of 50ml conc. HCl and 300ml distilled water. Dilute to 500ml with water. Stable.
- (3) N-(1-naphthyl)-ethylenediamine-dihydrochloride solution: Dissolve 500mg NED dihydrochloride in 500ml of water. Store in the dark. Replace monthly or as soon as a brownish color develops in the solution.
- (4) Hydrochloric acid: HCl, 1 + 3.
- (5) Sodium oxalate: 3.350g in 1000ml water.
- (6) Ferrous ammonium sulfate: 19.607g plus 20ml conc. H_2SO_4 dilute to 1L with dl water. This has to be standardized daily with a potassium chromate solution and ferroin indicator.
- (7) Stock nitrite solution: 1.232g of NaNO_2 in water and dilute to 1000ml. 1.00ml = 250 micrograms.
- (8) Standardize stock solution: Pipette in order: 50.00ml std. 0.05N KMnO_4 , 5ml conc. H_2SO_4 , and 50.0ml stock NO_2 . Shake gently and warm to 70-80°C. Discharge permanganate color with standard FAS solution in 10ml portions. Titrate excess with 0.05M KMnO_4 to a faint pink endpoint. Carry a water blank through the entire procedure.

$$A = \text{content of stock solution} = \frac{[(B \times C) - (D \times E)] \times 7}{F}$$

A = mg total NO₂⁻ in the stock solution

B = total ml standard KMnO₄ used

C = normality of standard KMnO₄

D = total ml standard reductant used, FAS

E = normality of standard reductant used, FAS

F = ml stock NaNO₂ solution taken for titration

Each 1.00ml of 00.05N KMnO₄ corresponds to 350µg NO₂⁻

(9) Intermediate nitrite solution: G = 12.5/A. Dilute G (approximately 50ml) to 250ml with water. 1ml = 50.0 µg N. Prepare daily.

(10) Standard nitrite solution: Dilute 10.00ml intermediate NO₂⁻ solution to 1000ml with water. 1.00ml = 0.500 micrograms N.

(11) 0.05N KMnO₄: 0.8g KMnO₄ per liter dl water. Keep in brown glass and age for one week. Decant supernate without disturbing sediment and standardize supernate with 0.05N ferrous ammonium sulfate solution.

(12) Ferroin indicator: dissolve 1.485g 1,10-phenanthroline monohydrate and 695mg FeSO₄ ·7H₂O in dl water and dilute to 100ml.

(13) FAS titrate: 0.25M. Dissolve 98g Fe(NH₄)₂(SO₄)₂·6H₂O in dl water. Add 20ml conc H₂SO₄, cool and dilute to 1000ml.

(14) Standard potassium dichromate: 0.0417M. Dissolve 12.259g K₂Cr₂O₇, primary standard grade, previously dried at 103°C for 2 hours, in dl water and dilute to 1L. Standardize K₂Cr₂O₇ daily by diluting 10ml of standard K₂Cr₂O₇ to 100ml. Add 30ml conc. H₂SO₄ and cool. Titrate with FAS titrate using 0.10 to 0.15ml (2 to 3 drops) of ferroin indicator.

Procedure

(1) Filter sample through a 0.45 µ membrane filter.

(2) To 50ml sample neutralized to pH 7.0, add 1ml sulfanilimide reagent and let it react for 2-8 minutes.

(3) Add 1.0ml NED dihydrochloride solution and mix immediately. Let stand at least 2 minutes but no more than 2 hours.

(4) Measure absorbance at 543nm.

Standard No.	Standards for Nitrite ml Standard Nitrite	Conc. (mg/l)
1	0	0
2	0.1	0.001
3	0.2	0.002
4	0.4	0.004
5	0.7	0.007
6	1.0	0.010
7	2.0	0.020

Calculations

$\text{mg NO}_2\text{-/L} = \frac{\text{g NO}_2\text{- (in 52ml final volume)}}{\text{ml sample}}$
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4. Ammonia, Ion sensitive Electrode

Method Number 417 E, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985, and, *Ammonia Combination Electrode*, Corning Glass Works Operating Instructions/Technical Specifications

Reagents

- (1) Ammonia-free water
- (2) Sodium Hydroxide, 6N. 240g of NaOH pellets in 1000mL of dl water. This is exothermic , allow solution to cool to adjust final volume to 1000mL.
- (3) Stock ammonium chloride solution, see Ammonia Nesslerization, reagent 4.
- (4) Standard ammonium chloride solution, see Ammonia Nesslerization, reagent 5.

Procedure

- (1) Prepare standards for Ammonia in a 125 erlenmeyer flask.

<u>Standards for Ammonia</u>		
Std. Number	ml Std. Ammonia	Concentration, ppm
1	0	0
2	0.2	0.049
3	0.5	0.122
4	1.0	0.244
5	2.5	0.610
6	4.0	0.976
7	10.0	2.400

- (2) Prepare Samples by pouring out 50mL into a clean 50mL erlenmeyer flask. Be sure to observe proper preservation procedure for the samples prior to analysis.
- (3) Equilibrate both samples and standards in the acid range with dilute HCl to pH 2. This is unnecessary for samples that have been preserved properly. Also make sure that both the samples and the standards are at the same temperature.
- (4) Connect the Ammonia ISE to its input connectors. With the pH Meter in standby turn the mode switch to -mv and begin calibration. Start on the blank, and add 1mL of 6N NaOH to bring the pH above 11. With stirring on a magnetic stirrer with a teflon stir bar zero the Meter to the blank. Then proceed to the standards of greater strength. Measure ammonia content as one adds the NaOH, any delay may cause the loss of Ammonia from the solution as soon as it is made basic. Always immerse the probe at an angle so that gas does not get trapped on the tip. 30° is usually sufficient. Do not stir the solution so violently that air is mixed in the solution. Always allow 2-3 minutes for the mv reading to stabilize.

- (4) Remove the electrode and rinse with a small amount of sample. Immerse the electrode into the sample with gentle stirring. Add 1 mL of 6N NaOH. Take a mv reading when the display is stable, usually 1-2 minutes.
- (5) Construct a curve from the standards on four-cycle semilog paper with concentration in ppm on the log axis and mv on the linear axis.
- (6) When not in use rinse the electrode in dl water and blot it dry. Immerse the electrode in 0.05M Ammonium Chloride. The membrane can be maintained in such a manner for 3-4 month.

5. Ammonia, Phenate Method

Method Number 417 C, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Ammonia Free Water
- (2) Hypochlorous acid reagent: To 40 mL of dl water add 10 mL 5% NaOCl solution obtained from commercial chlorox bleach. Adjust to pH 6.5-7.0 with HCl. Reagent is only stable for one week.
- (3) Manganous Sulfate Solution, 0.006N: Dissolve 50 mg $MnSO_4 \cdot H_2O$ in 100mL dl water.
- (4) Phenate Reagent: Dissolve 2.5g NaOH and 10g phenol in 100mL dl water. This reagent is only stable for one week. Handle Phenol with care.
- (5) Stock ammonium chloride solution, see Ammonia Nesslerization, reagent 4.
- (6) Standard ammonium chloride solution, see Ammonia Nesslerization, reagent 5.

Procedure

- (1) 10 mL sample in a 50 mL erlenmeyer.
- (2) Add one drop of Manganous Sulfate Solution.
- (3) Place on a magnetic stirrer.
- (4) Add 0.5 mL of Hypochlorous Acid Solution.
- (5) Immediately add 0.6 mL of Phenate Reagent, one drop at a time.
- (6) Carry a blank and standards through the entire procedure

<u>Standards for Ammonia</u>		
Std. Number	ml Std. Ammonia	Concentration, ppm
1	0	0
2	0.2	0.049
3	0.5	0.122
4	1.0	0.244
5	2.5	0.610
6	4.0	0.976
7	10.0	2.400

- (7) Measure absorbance at 630 nm. Color development is stable after 10 minutes, and to 24 hours.

6. Ammonia, Nesslerization

Method Number 417 B, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Zinc sulfate solution: dissolve 100g $ZnSO_4 \cdot 7H_2O$ and dilute to 1L with ammonia-free water.
- (2) EDTA stabilizer: dissolve 50g of disodium ethylenediamine tetracetate dihydrate in 60ml of ammonia-free water containing 10g of NaOH, heat if necessary. Cool and dilute to 100 ml.
- (3) Nessler reagent: Dissolve 100g HgI_2 and 70g KI in a small amount of ammonia-free water. Add slowly, with stirring, to a solution of 160g NaOH in 500ml water. Dilute to 1L. Keep out of sunlight. Reagent keeps up to a year and can be checked by development of the characteristic color with 0.1mg NH_3-N/L within 10 min. without the development of a precipitate within 2 hours.
- (4) Stock ammonia solution: Dissolve 3.819g anhydrous NH_3Cl (dry at 100°C) in ammonia-free water, and dilute to 1L. 1.00ml = 1.00mg N = 1.22mg NH_3 .
- (5) Standard ammonium solution: Dilute 10.00ml no. 4 to 1L with pure water. 1.00ml = 10.00 μ g N = 12.2 μ g NH_3 .
- (6) Borate buffer: Add 88ml 0.1N NaOH solution to 500ml of 0.025M sodium tetraborate solution (9.5g $N_3B_4O_7 \cdot 10H_2O/L$) and dilute to 1L.
- (7) Sodium hydroxide, 6N: Dissolve 240g NaOH in water and dilute to 1L.

Procedure

- (1) If interferences are noted, remove residual chlorine from the freely collected sample, do not store chlorinated samples without prior dechlorination. Add 1ml $ZnSO_4$ solution to 100ml sample and mix. Add 0.4 to 0.5ml 6N NaOH to obtain a pH of 10.5. Mix and let sample stand for a few minutes. A heavy, flocculent precipitate should form, leaving the supernate clear and colorless. Filter or centrifuge sample. Pretest any filter paper with some nessler reagent to make sure it contains no ammonia. Filter sample and discard the first 25ml of filtrate. Samples containing more than 10mg NH_3-N/L may lose ammonia during treatment. Dilute such a sample to the sensitive range for renesslerization before treatment.
- (2) Use 50ml sample or a portion diluted to 50ml. If the sample contains high concentrations of ions like calcium and magnesium that cause turbidity, or precipitate with the nessler reagent, add 1 drop of EDTA reagent. Mix. Add 2ml of nessler reagent.
- (3) Mix samples by swirling at least six times. Keep temperature and reaction time constant in blank, samples and standards. Let the reaction proceed at least 10 minutes after addition of the nessler reagent. If color development is weak use a 30 minute contact time.
- (4) Measure the absorbance at 425nm.

CHOCTAWHATCHEE - S. O. P.

Date 12/13/89

Page Number 24

<u>Standards for Ammonia</u>		
Std. Number	ml Std. Ammonia	Concentration, ppm
1	0	0
2	0.2	0.049
3	0.5	0.122
4	1.0	0.244
5	2.5	0.610
6	4.0	0.976
7	10.0	2.400

Detection Limit = 1 μ g Ammonia

7. Phosphates, filtration

Filter samples through a 0.45 μ membrane filter. Glass fiber filters may be used to prefilter sample. Wash membranes by soaking in dl water to remove any residual phosphates. To wash, soak 50 filters in 2L dl water for 1 hour. Change water and soak for additional 3 hours.

filtration step: separates total and dissolved phosphate analysis

8. Phosphates, condensed phosphates

Method Number 424 B, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Phenolphthalein: aqueous solution
- (2) Strong acid: add 300ml of conc. H_2SO_4 to about 600ml dl water. When cool add 4.0ml conc. HNO_3 , dilute to 1L.
- (3) Sodium hydroxide: NaOH 6N, see Ammonia, Ion Sensitive Electrode, reagent 2.

Procedure

- (1) 50 ml sample
- (2) Add 2 drops phenolphthalein, and acidify dropwise with the strong acid solution if a red color ensues. Add another ml of acid after discharge of color to each sample.
- (3) Boil gently for 90 minutes, being careful to keep sample volume at around 25-50ml, or autoclave for 30 minutes at 98-137kPa with foil caps on the flasks. Cool and neutralize to a faint pink color with 6N NaOH. Restore to original 100ml volume with dl water.
- (4) Run the phosphate standards through step number 3 to assure a good calibration curve.
- (5) Colorimetric method: run ascorbic acid test to quantitate.

9. Phosphates, total phosphate

Method Number 420 C, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Sulfuric acid: conc.
- (2) Nitric acid: conc.
- (3) Phenolphthalein: aqueous solution
- (4) Sodium hydroxide: 1N NaOH

Procedure

- (1) Add 50ml sample to a micro-kjeldahl flask. Add 1ml conc. H_2SO_4 and 5ml conc HNO_3 .
- (2) Digest to a volume of 1ml. Continue digestion to a discharge of color which signifies the removal of HNO_3 , approximately 40 minutes.
- (3) Cool. Add about 20ml dl water.
- (4) Add 1 drop of phenolphthalein and as much 1N NaOH as necessary to produce a faint pink tinge.
- (5) Transfer to erlenmeyer flask, with filtration (only if necessary), and adjust sample volume to 100ml with dl water.
- (6) Colorimetric Method, run ascorbic acid.

10. Ortho-phosphates, ascorbic acid method

Method Number 424 F, *Standard Methods for the Examination of Water and Wastewater*, Sixteenth Edition, 1985.

Reagents

- (1) Sulfuric acid: 70ml conc. H_2SO_4 in 500ml water (5N), or 280ml in 2L.
- (2) Potassium antimonyl tartrate solution: 1.3715g in 400ml water, and dilute to 500ml. Store in glass stoppered bottle.

CHOCTAWHATCHEE - S. O. P.

Date 12/13/89

Page Number 27

(3) Ammonium molybdate solution: 20g in 500ml water. Store in glass stoppered bottle.

(4) Ascorbic acid (0.01 M): 1.76g in 100ml dl water; stable for 1 week at 4°C, or 2.64g in 150ml dl water, or 5.26g in 300ml dl water.

(5) Combined reagent:

[100ml total]	[500ml total]	[1000ml total]
50ml 5N H ₂ SO ₄	250ml H ₂ SO ₄	500ml H ₂ SO ₄
5ml PAT	25ml PAT	50ml PAT
15ml AM	75ml AM	150ml AM 30ml
AA 150ml AA	300ml	

(6) Stock phosphate solution: 219.5mg anhydrous KH₂PO₄ in 1000ml water 1ml = 50 µg PO₄

(7) Standard phosphate solution: dilute 50ml stock phosphate solution to 1000ml with water. 1ml = 2.5 microgram PO₄

Procedure

- (1) pour 50ml sample into a 125ml erlenmeyer flask
- (2) add 2 drop phenolphthalein indicator
- (3) if red color develops add 5N H₂SO₄ dropwise until discharge of color
- (4) add 8ml of the combined reagent and mix thoroughly
- (5) after 10 minutes, but not before 30 minutes, measure absorbance at 880nm. Use a reagent blank as the reference.

Standards for Phosphate		
Standard No.	ml Standard Phosphate	Conc. (mg/l)
1	0	0
2	0.2	0.010
3	0.5	0.025
4	1.0	0.050
5	5.0	0.250
6	10.0	0.500
7	20.0	1.000

Detection Limit = 10 µg Phosphate

V. Solutions

1. Lugal's Solution, for preserving plankton samples

20g KI	60g KI
10g I, crystalline	30gI, crystalline
200ml dl H ₂ O	600ml dl H ₂ O
20ml Acetic Acid, glacial	60ml Acetic Acid, glacial

Note: Mix the dl water with the glacial acetic acid first, then add the other ingredients with stirring.

Dose:

0.3ml of Lugal's will preserve 100ml of sample if it is stored in the dark. For long term storage use 0.7ml of Lugal's for 100ml of sample.

2. Bouin's Solution, for preserving histological samples

10 parts formalin
90 parts picric acid
1 part 1+1 acetic acid

Note: The solubility of picric acid in water is 1g/78ml or 12g/L. Prepare the solution with care. Dry crystalline picric acid is explosive when subjected to any impact, such as dropping it on the floor. This solution will fix the tissues of ones hand upon contact.

3. Probe Juice, for Y.S.I. D.O. Probes

Use half saturated KCl in dl water. A saturated KCl solution is used with the Calomile Electrodes. Dilute the saturated solution with one part water to get a half saturated solution.

4. Probe Cleaner, for the Y.S.I. S.C.T. Probes

Any commercial foaming tile cleaner can be used, but it is best to use a home brew cleaner of : 10 parts dl water; 10 parts isopropanol; and 1 part concentrated HCl.

5. Probe Cleaner, for the Y.S.I. D.O. Probes

To clean the gold electrode rub its' surface with an abrasive eraser like those used on ball point ink pens. To clean the silver electrode empty out the probe juice and fill the chamber with 14% NH₄OH. Soak for 10 minutes. Rinse the chamber with several volumes of dl water. Fill chamber with probe juice, and replace membrane.

Appendix III

Photographic Atlas of Dominant Phytoplankton Species found in the Choctawhatchee Bay System

A note on Photomicrographs and legends

The present atlas is the continuation of the previously prepared "An Atlas of diatoms and other forms from selected Drainage areas in Central and North Florida" (Prasad & Livingston 1987), and includes only those forms that occur in brackishwater and marine environments. This atlas is a photographic survey at the light microscopic level of over 255 algal taxa belonging to 90 genera. The groups represented here include Bacillariophyceae (Diatoms), Chrysophyceae (Golden-brown algae), Cyanophyceae (Blue-green algae), and Dinophyceae (Dinoflagellates). The freshwater atlas mentioned above contains illustrations for over 460 taxa belonging to 72 genera. These two works, together, provide a comprehensive account of the algal species present in river-bay systems

The micrographs were taken with two NIKON research microscopes, one (LABOPHOT) being equipped for bright and phase contrast illumination, the other (MICROPHOT) exclusively for Nomarski Differential Interference Contrast (DIC). Most of the photographs were taken with oil-immersion phase contrast objectives (100x with 1.25 numerical aperture and 0.16 mm working distance). The DIC was used in cases where valves lying on top of one another had to be separated optically. For e.g. in separating valves of *Achnanthes*, *Cocconeis*, *Mastogloia* etc. All micrographs were taken on Pan-X black and white film and the prints and enlargements were made on Kodak Poly contrast III RC multigrade photographic paper.

The arrangement of photomicrographs in this atlas is quite simple. The plates are arranged in different phyla or divisions of generally accepted systems of classification. The genera and species in each family represented are not arranged in any particular order. Each taxon is illustrated by more than one figure reflecting the range of variability from each locality and season. Many species of diatoms that are weakly silicified such as those of *Chaetoceros* and *Rhizosolenia* were mounted in distilled water without cleaning them with acids. The legends for plates are kept brief. Location and month of collection are included as notes regarding the spacial and temporal occurrence of each taxon, followed by the numerical magnification.

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Register of Taxa Illustrated

Name of the Taxa	Plate Number
<i>Achnanthes citronella</i>	34
<i>Achnanthes mannifera</i>	33
<i>Actinocyclus curvatulus</i>	10
<i>Actinocyclus ehrenbergii</i>	10, 20, 46
<i>Actinocyclus ehrenbergii</i> v. <i>crassa</i>	10
<i>Actinocyclus ehrenbergii</i> v. <i>tenella</i>	10
<i>Actinocyclus</i> sp.	10
<i>Actinoptychus senarius</i>	10
<i>Actinoptychus splendens</i>	11
<i>Amphiprora alata</i>	38
<i>Amphora arenaria</i>	41
<i>Amphora astrearia</i>	41
<i>Amphora binoides</i> v. <i>interrupta</i>	39
<i>Amphora clara</i>	33
<i>Amphora clevei</i>	33
<i>Amphora coffeaeformis</i>	33, 41
<i>Amphora decussata</i>	41
<i>Amphora</i> cf. <i>exornata</i>	33
<i>Amphora</i> cf. <i>graeffi</i> v. <i>minor</i>	41
<i>Amphora holsatica</i>	39
<i>Amphora laevis</i>	33
<i>Amphora ocellata</i>	33, 41
<i>Amphora proteus</i>	33
<i>Amphora</i> sp.	39, 41

<i>Asteromphalus flabellatus</i>	10
<i>Asterionella japonica</i>	31, 32
<i>Attheya decora</i>	25
<i>Aulacodiscus argus</i>	27
<i>Azpeitia nodulifer</i>	10
<i>Bacillaria paxillifer</i>	32, 44, 45
<i>Bacteriatrum delicatulum</i>	21
<i>Bacteriastrum hyalinum</i>	9, 21
<i>Biddulphia mobiliensis</i>	26
<i>Biddulphia regia</i>	27
<i>Biddulphia sinensis</i>	26
<i>Caloneis oregonica</i>	33
<i>Campylodiscus clypeus</i>	43
<i>Campylodiscus echeneis</i>	43
<i>Campylodiscus limbatus</i>	33
<i>Campyloneis grevillei</i>	34
<i>Ceratium belone</i>	4
<i>Ceratium furca</i>	4
<i>Ceratium fusus</i>	4
<i>Ceratium hircus</i>	23
<i>Ceratium pentagonum</i>	4
<i>Ceratium teres</i>	4
<i>Ceratium trichoceros</i>	2, 4
<i>Ceratium tripos</i>	1, 3
<i>Ceratium tripos</i> v. <i>atlanticum</i>	2
<i>Chaetoceros affinis</i>	21, 22, 24
<i>Chaetoceros borealis</i>	22

Chaetoceros brevis	22
Chaetoceros compressa	21, 23, 24
Chaetoceros curvisetus	21
Chaetoceros decipiens	20, 21, 22
Chaetoceros dichæta	23
Chaetoceros didymus	23
Chaetoceros didymus v. protuberance	23
Chaetoceros diversus	20
Chaetoceros eibenii	24
Chaetoceros lorenzianus	21, 22
Chaetoceros messanensis	20
Chaetoceros peruvianus	23, 27
Chaetoceros pseudocurvisetus	20, 21, 22
Chatoceros socialis	20
Chaetoceros subsecundes	21, 22
Chaetoceros tere	21, 24
Cladopyxis brachiolata	7
Climacodium frauenfeldianum	25
Cocconeis decipiens	34
Corethron criophilum	10
Coscinodiscus apiculatus	3, 11, 13
Coscinodiscus asteromphalus	14
Coscinodiscus centralis	11, 12, 13
Coscinodiscus curvatulus	12, 14
Coscinodiscus cf. gigas	12
Coscinodiscus granii	15
Coscinodiscus jonesianus	14, 20

<i>Coscinodiscus perforatus</i> v. <i>pavillardii</i>	15
<i>Coscinodiscus oculus-iridis</i>	11,13
<i>Cymbella</i> sp.	39
<i>Cyclotella choctawhatchiensis</i>	8
<i>Cyclotella cryptica</i>	8
<i>Cyclotella meneghiniana</i>	8
<i>Cyclotella striata</i>	8
<i>Cymatosira belgica</i>	26
<i>Cymatosira lorenziana</i>	26,27
<i>Delphineis livingstonii</i>	30
<i>Delphineis surirella</i>	29,30
<i>Denticula</i> cf. <i>kuetzingii</i>	45
<i>Denticula confervacea</i>	16
<i>Dictyoneis marginatus</i>	34
<i>Dimeregramma fulvum</i>	30
<i>Dimeregramma furcigerum</i>	30
<i>Dimeregramma marinum</i>	30
<i>Dimeregramma minor</i>	30
<i>Dinobryon sertularia</i>	1
<i>Dinophysis caudata</i>	5,7
<i>Dinophysis caudata</i> v. <i>acutiformis</i>	40
<i>Dinophysis caudata</i> v. <i>pedunculata</i>	5
<i>Dinophysis</i> spp.	5
<i>Diploneis crabro</i>	37
<i>Diploneis parma</i>	33
<i>Diploneis reichardtii</i>	38
<i>Diploneis</i> cf. <i>weissflogii</i>	38

<i>Diplopsalis lenticula</i>	7
<i>Ditylum brightwelli</i>	26
<i>Eucampia cornuta</i>	28
<i>Eucampia zoodiacus</i>	28
<i>Eunatogramma laeve</i>	26,27
<i>Eupodiscus radiatus</i>	12,27
<i>Exuviella baltica</i>	7,40
<i>Fragilaria leptostauron</i> v. <i>rhomboides</i>	31
<i>Grammatophora oceanica</i>	29
<i>Guinardia flaccida</i>	16
<i>Gymnodinium rotundatum</i>	7
<i>Gyrosigma fasciola</i>	37
<i>Gyrosigma fasciola</i> v. <i>arcuata</i>	38
<i>Hantzschia</i> sp.	45
<i>Haslea</i> sp.	37
<i>Hemiaulus hauckii</i>	25
<i>Hemiaulus membranaceus</i>	25,28
<i>Hemiaulus sinensis</i>	25
<i>Hemidiscus cuneiformis</i>	10
<i>Hyalodiscus radiatus</i>	8
<i>Lauderia borealis</i>	9,16
<i>Leptocylindrus danicus</i>	10
<i>Licmophora abbreviata</i>	31
<i>Lithodesmium undulatum</i>	27
<i>Mastogloia angulata</i>	36
<i>Mastogloia baldjikiana</i>	34,35,38
<i>Mastogloia braunii</i>	35

<i>Mastogloia elegans</i>	35
<i>Mastogloia elliptica</i>	34, 35
<i>Mastogloia erythraea</i>	35
<i>Mastogloia euxina</i>	39
<i>Mastogloia foliolum</i>	37, 38
<i>Mastogloia ignorata</i>	34
<i>Mastogloiaportierana</i>	36
<i>Mastogloia smithii</i>	36
<i>Mastogloia</i> sp.	36
<i>Mastogloia subacuta</i>	34
<i>Mastoneis biformis</i>	36
<i>Melosira undulata</i>	8, 12
<i>Microcoleus lyngbyaceus</i>	1
<i>Navicula abunda</i>	35, 37, 40
<i>Navicula</i> cf. <i>algida</i>	40
<i>Navicula clamans</i>	39
<i>Navicula clavata</i>	38
<i>Navicula cruciculoides</i>	38
<i>Navicula diffluens</i>	35
<i>Navicula directa</i>	39
<i>Navicula finmarchica</i>	33
<i>Navicula forcipata</i>	31, 33, 38
<i>Navicula granulata</i>	36, 40
<i>Navicula lyra</i>	37, 37
<i>Navicula lyra</i> v. <i>insignis</i>	35
<i>Navicula maculosa</i>	38
<i>Navicula marina</i>	36

Navicula menaiana	35
Navicula normalis	37
Navicula cf. nummularia	39
Navicula peregrina	37
Navicula pygmaea	36.
Navicula cf. pseudoscutiformis	36
Navicula spp.	37, 39, 40
Navicula spectabilis	38
Navicula cf. tuscula	40
Navicula yarrensis v. americana	40
Neodelphineis pelagica	30
Nitzschia closterium	41
Nitzschia cicumsuta	41, 44
Nitzschia constricta	42
Nitzschia dissipata	45
Nitzschia dubia	45
Nitzschia epithemoides	42
Nitzschia fossilis	42
Nitzschia fusoides	43
Nitzschia hungarica	41
Nitzschia cf. hybrida	42
Nitzschia lacunaris	45
Nitzschia lanceolata	42
Nitzschia longissima	41, 43
Nitzschia marginulata	42
Nitzschia normanii	44
Nitzschia obtusa	42

<i>Nitzschia pellucida</i>	42
<i>Nitzschia pungens</i> v. <i>atlantica</i>	42, 43
<i>Nitzschia reversa</i>	41, 42
<i>Nitzschia scalaris</i>	43
<i>Nitzschia seriata</i>	43
<i>Nitzschia sigma</i>	43, 44, 45
<i>Nitzschia</i> cf. <i>vitrea</i>	42
<i>Odontella rhombosa</i>	27
<i>Opephora martyi</i>	29
<i>Opephora pacifica</i>	30
<i>Ornithoceros magnificus</i>	7
<i>Oxytoxum</i> so.	5
<i>Perisonnoe crucifera</i>	30
<i>Peridinium conicum</i>	6
<i>Peridinium depressum</i>	3
<i>Peridinium divarigatum</i>	3
<i>Peridinium divergens</i>	6
<i>Peridinium nipponicum</i>	6
<i>Peridinium oblongum</i>	6
<i>Peridinium pallidum</i>	6
<i>Peridinium venustum</i>	6
<i>Phalacroma cuneus</i>	7
<i>Plagigramma staurophorum</i>	30, 32
<i>Pleurisigma strigosum</i>	37
<i>Podosira hormoides</i>	13
<i>Podosira stelliger</i>	8
<i>Porosira</i> cf. <i>glacialis</i>	10

<i>Prorocentrum gracile</i>	7
<i>Prorocentrum micans</i>	7
<i>Prorocentrum minimum</i>	7
<i>Pseudauliscus radiatus</i>	27
<i>Rhabdonema adriaticum</i>	29
<i>Rhaphoneis amphiceros</i> v. <i>gemma</i>	30
<i>Rhizosolenia alata</i>	17,19
<i>Rhizosolenia calcaravis</i>	3,17
<i>Rhizosolenia castracanei</i>	17,19
<i>Rhizosolenia fragilissima</i>	16,18
<i>Rhizosolenia imbricata</i>	3,17,19
<i>Rhizosolenia robusta</i>	17,18
<i>Rhizosolenia setigera</i>	18
<i>Rhizosolenia stolterfothii</i>	16,17
<i>Rhizosolenia styliformis</i>	18,19
<i>Skeletonema costatum</i>	8
<i>Stellerima microtrias</i>	15
<i>Stephanopyxis pameriana</i>	8
<i>Stephanopyxis turris</i>	8,40
<i>Striatella interrupta</i>	16
<i>Striatella unipunctata</i>	29,31
<i>Surirella amphioxysis</i>	46
<i>Surirella biseriata</i>	46
<i>Surirellafastuosa</i>	43,46
<i>Surirella febigeri</i>	44
<i>Surirella gemma</i>	46
<i>Surirella linearis</i>	44,46

<i>Surirella robusta</i>	43
<i>Synedra pulchella</i>	31
<i>Synedra tabulata</i>	31
<i>Thalassionema nitzschioides</i>	30, 32
<i>Thalassiosira decipiens</i>	9
<i>Thalassiosira eccentrica</i>	9
<i>Thalassiosira lineatus</i>	9
<i>Thalassiosira oestrupii</i>	9
<i>Thalassiosira tumida</i>	9
<i>Thalassiothrix frauenfeldii</i>	32
<i>Thalassiothrix longissima</i>	31, 32
<i>Thalassiothrix mediterranea v. pacifica</i>	31
<i>Trichodesmium erythraeum</i>	1
<i>Trigonium arcticum</i>	40
<i>Trigonium alternans</i>	26, 27

Plate 01

- Fig. 1: *Microcoleus lyngbyaceus* (Kuetz.) Crouan.
Choctawhatchee Bay, north Florida, Station C-3.
10/87. x75.
- Fig. 2: *Trichodesmium erythraeum* (Ehr.) Gomont.
Choctawhatchee Bay, north Florida, Station C-3.
10/87. x75.
- Fig. 3: *Trichodesmium erythraeum* (Ehr.) Gomont.
Choctawhatchee Bay, north Florida, Station C-3.
10/87. x75.
- Fig. 4: *Ceratium tripos* Schroeder
Choctawhatchee Bay, north Florida, Station 22.
9/85. x500.
- Fig. 5: *Dinobryon sertularia* Ehr.
Choctawhatchee Bay, north Florida, Station 31.
12/85. x350.
- Fig. 6: *Ceratium tripos* (O. F. Mueller) Nitzsch.
Choctawhatchee Bay, north Florida, Station 3
5/86. x150.
- Fig. 7: *Ceratium tripos* (O. F. Mueller) Nitzsch.
Choctawhatchee Bay, north Florida, Station 3.
5/86. x250.
- Fig. 8: *Ceratium tripos* (O. F. Mueller) Nitzsch.
Choctawhatchee Bay, north Florida, Station 3.
5/86. x250.
- Fig. 9: *Nitzschia longissima* (Breb.) Ralfs.
Choctawhatchee Bay, north Florida, Station 15.
10/87. x250.

PLATE 1

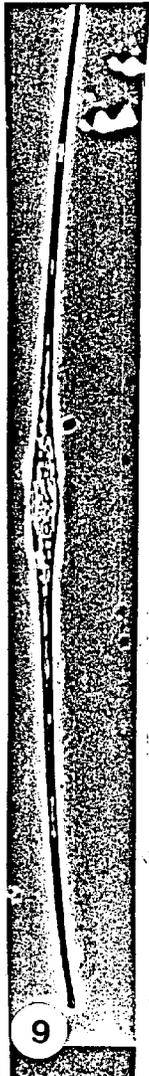
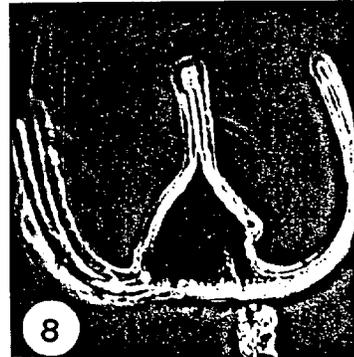
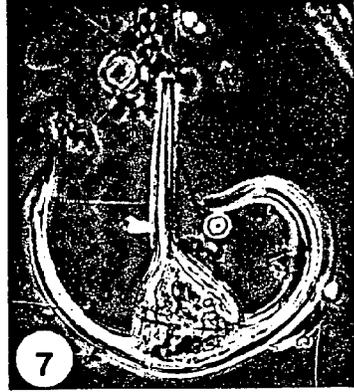
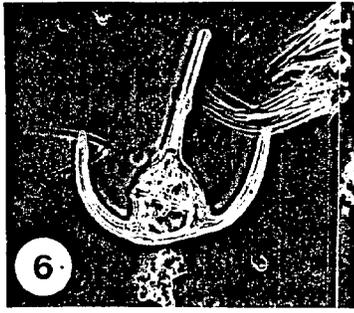
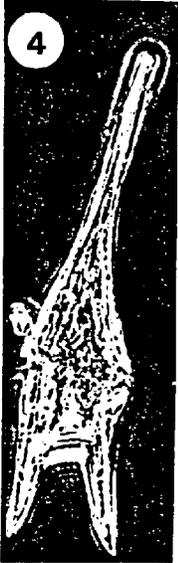
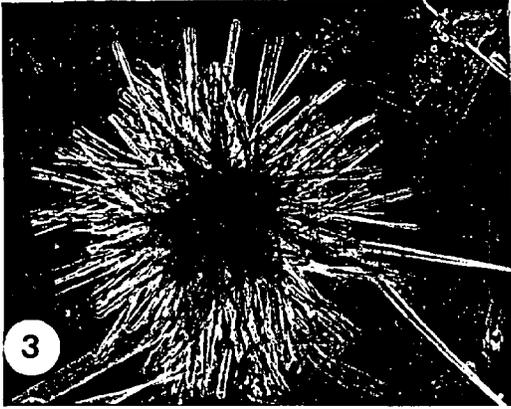
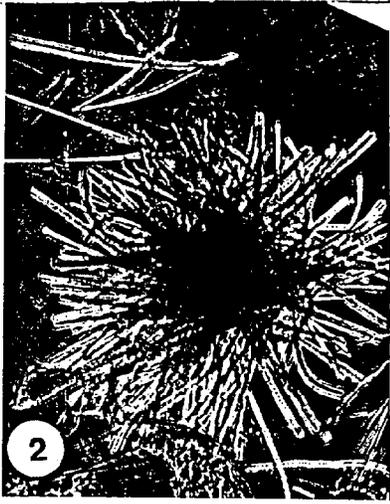


Plate 02

- Fig. 1: *Ceratium trichoceros* (Ehr.) Kofoid.
Choctawhatchee Bay, north Florida, Station C-3.
10/87. x100.
- Fig. 2: *Ceratium tripos* v. *atlanticum* Ostenfeld.
Choctawhatchee Bay, north Florida, Station 31.
3/86. x500.
- Fig. 3: *Ceratium hircus* Schroeder.
Choctawhatchee Bay, north Florida, Station 31.
11/85. x500.
- Figs.4,5: *Ceratium tripos* (Mueller) Nitzsch.
Choctawhatchee Bay, north Florida, Station 15.
7/87. x500.
- Fig. 6: *Ceratium hircus* Schroeder.
Choctawhatchee bay, north Florida, Station 03.
10/85. x500.
- Fig. 7: *Ceratium hircus* Schroeder
Choctawhatchee bay, north Florida, Station 15.
7/87. x250.
- Figs.8,9 : *Ceratium tripos* (Mueller) Nitzsch.
Choctawhatchee Bay, north Florida, Station 15.
7/87. x500.
- Fig. 10: *Ceratium tripos* (Mueller) Nitzsch.
FSU Marine Lab., north Florida, Station M-4.
2/88. x500.
- Fig. 11: *Ceratium hircus* Schroeder.
Choctawhatchee Bay, north Florida, Station 15.

PLATE 2

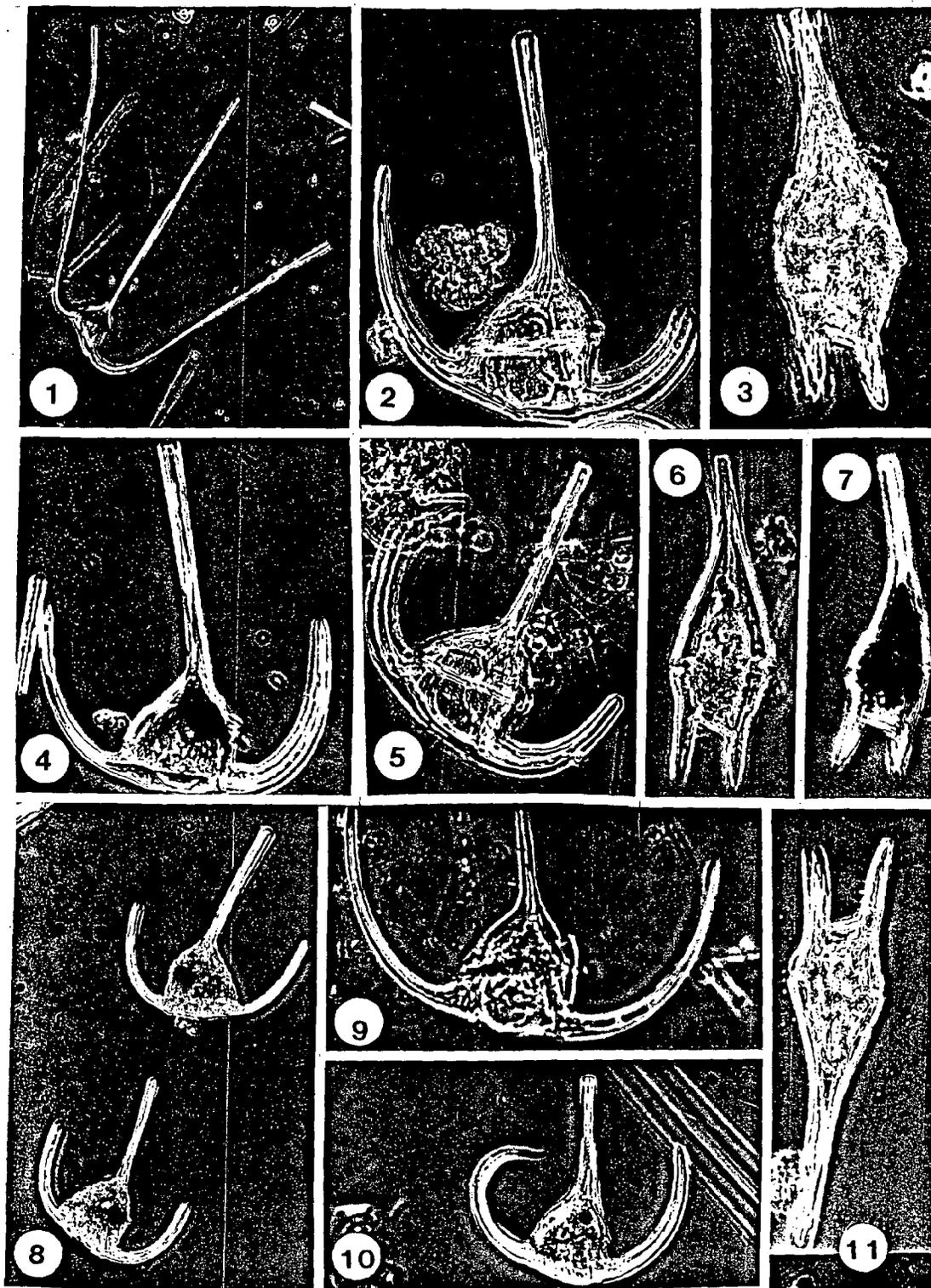


Plate 03

- Figs. 1,2: *Peridinium depressum* Bailey.
Choctawhatchee Bay, north Florida, Station 34.
9/85. x500.
- Figs. 3,4: *Ceratium hircus* Schroeder.
Choctawhatchee Bay, north Florida, Station 15.
1/86. x500.
- Fig. 5: *Ceratium tripos* (Mueller) Nitzsch.
Choctawhatchee Bay, north Florida, Station 34.
3/86. x500.
- Fig. 6: *Rhizosolenia imbricata* Brightw.
Choctawhatchee Bay, north Florida, Station 34.
3/86. x150.
- Fig. 7: *Peridinium divarigatum* Meunier
Choctawhatchee Bay, north Florida, Station 15.
1/86. x500.
- Fig. 8. *Ceratium tripos* (O. F. Mueller) Nitzsch.
Choctawhatchee Bay, north Florida, Station 11.
5/86. x400.
- Fig. 9: *Coscinodiscus apiculatus* Ehr.
Apalachicola Bay, north Florida, Station E-4.
2/88. x500.
- Figs. 10,11: *Rhizosolenia calcar-avis* Schulze.
FSU Marine Lab., north Florida. 12/86. x100.

PLATE 3

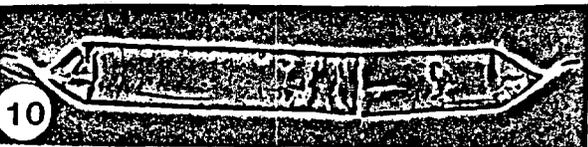
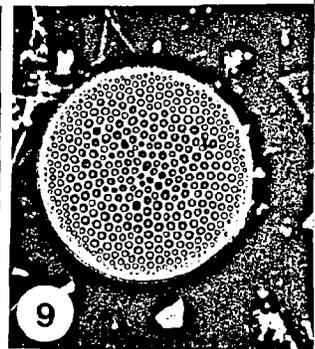
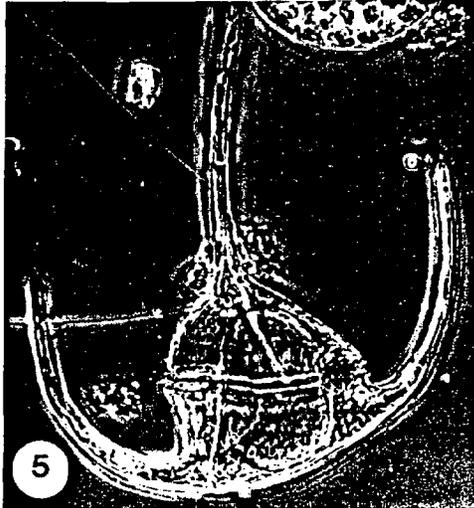
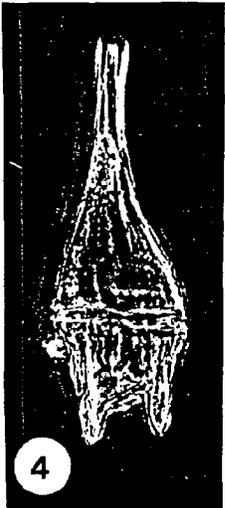
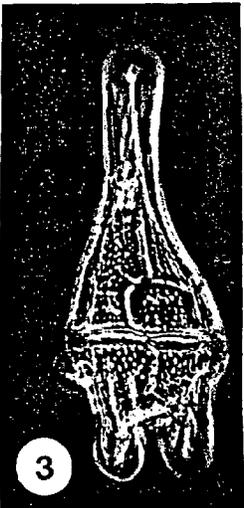
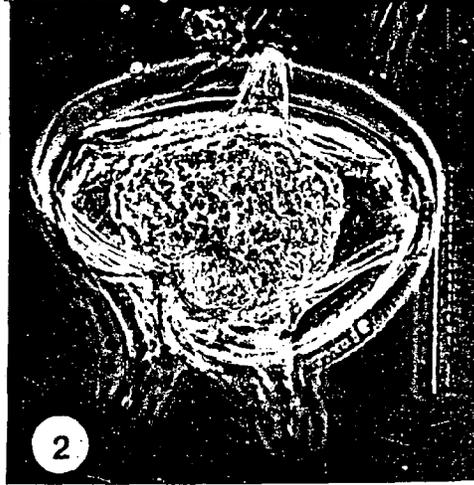
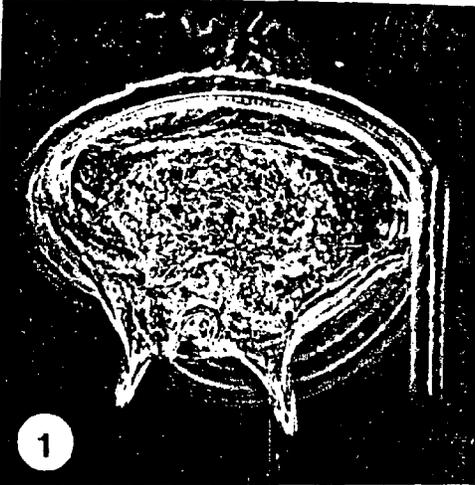


Plate 04

- Fig. 1: *Ceratium belone* Cleve.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.
- Fig. 2: *Ceratium fusus* (Ehr.) Dujardin
Choctawhatchee Bay, north Florida, Station 15.
7/87. x200.
- Fig. 3: *Ceratium fusus* (Ehr.) Dujardin.
Choctawhatchee Bay, north Florida, Station 07.
7/87. x200.
- Fig. 4: *Ceratium fusus* (Ehr.) Dujardin.
Choctawhatchee Bay, north Florida, Station 15.
7/87. x200.
- Fig. 5: *Ceratium furca* (Ehr.) Claparede.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.
- Fig. 6: *Ceratium teres* Kofoid.
Choctawhatchee Bay, north Florida, Station 07.
7/87. x500.
- Fig. 7: *Ceratium hircus* Schroeder
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.
- Fig. 8: *Ceratium trichoceros* (Ehr.) Kofoid.
Choctawhatchee Bay, north Florida, Station 03.
3/87. x500.
- Fig. 9: *Ceratium pentagonum* Gourret.
Apalachee Bay, north Florida, Station A-3.
10/87. x500.

PLATE 4

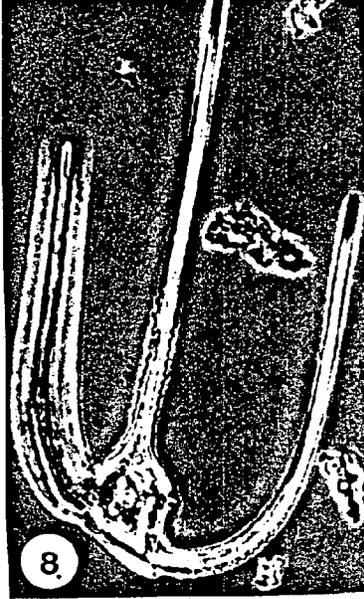
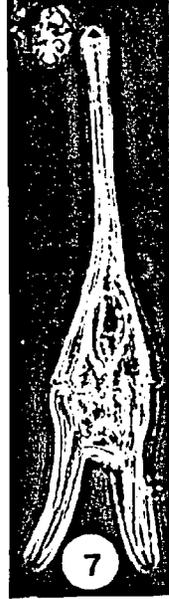
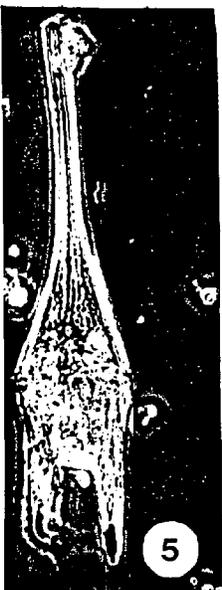
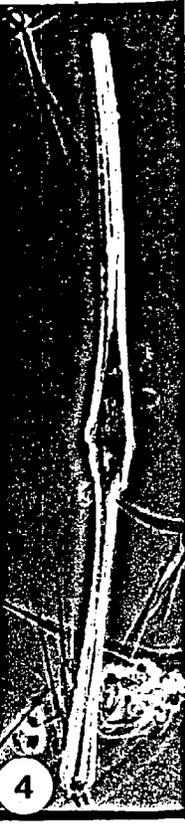
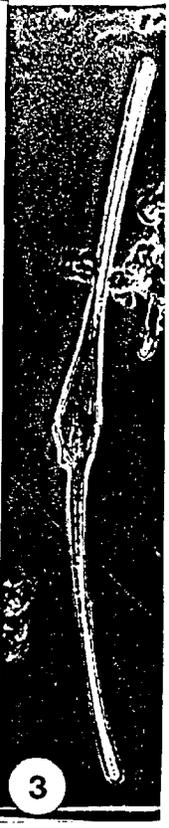
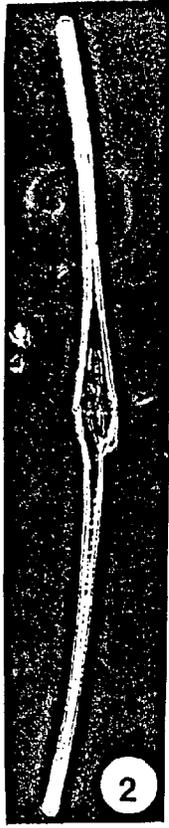


Plate 05

- Figs. 1,2: *Dinophysis caudata* v. *pedunculata* Schmidt.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 3: *Dinophysis caudata* v. *pedunculata* Schmidt.
Apalachee Bay, north Florida, Station A-1.
2/88. x500.
- Figs. 4,5: *Dinophysis caudata* v. *pedunculata* Schmidt.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 6: *Oxytoxum* sp.
Apalachee Bay, north Florida, Station A-1.
2/88. x500.
- Figs 7,8: *Dinophysis caudata* Savile-Kent.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 9: *Dinophysis* sp.
Choctawhatchee Bay, north Florida, Station 34.
11/85. x500.
- Fig. 10: *Dinophysis caudata* v. *pedunculata* Schmidt.
Apalachee Bay, north Florida, Station A-1.
2/88. x500.
- Fig. 11: *Dinophysis* ? sp.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 12: *Dinophysis caudata* Savile-Kent.
Choctawhatchee bay, north Florida, Station 38.
10/85. x500.

PLATE 5

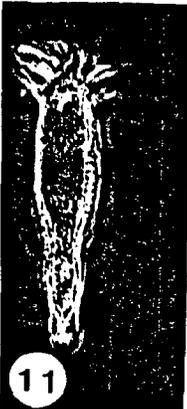
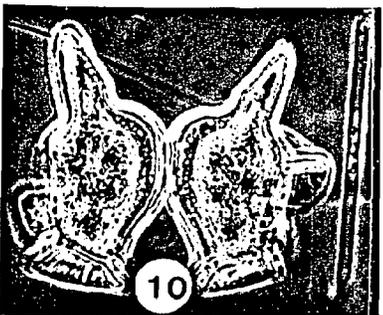
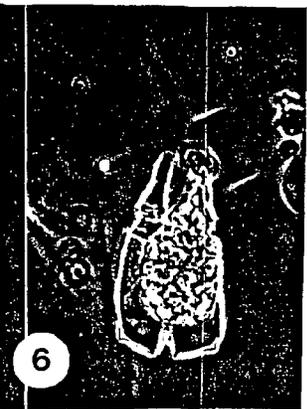
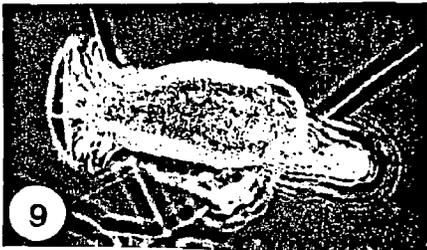
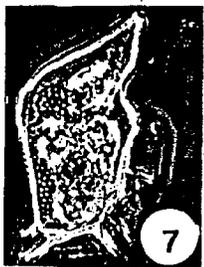
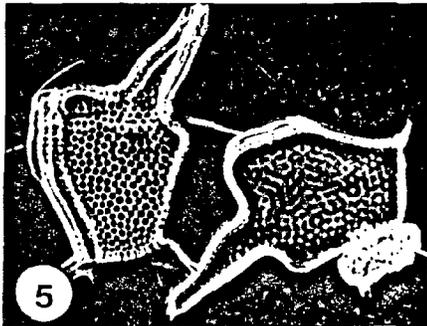
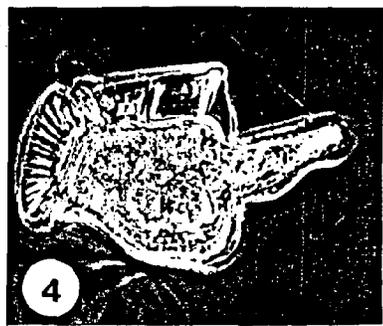
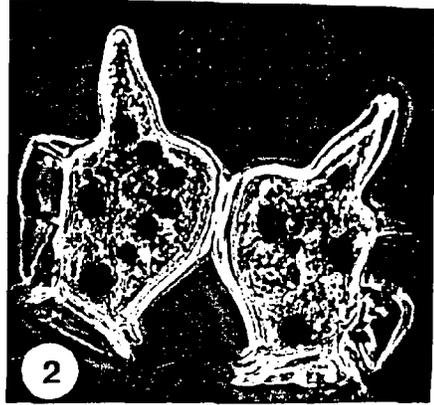
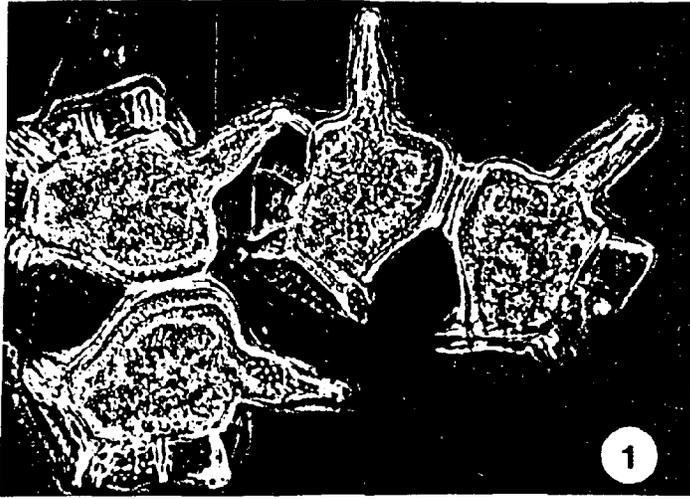


Plate 06

- Fig. 1: *Peridinium divergens* Ehr.
Choctawhatchee Bay, north Florida, Station 31.
7/87. x300.
- Fig. 2: *Peridinium conicum* (Grand) Ostenfeld & Schmidt.
FSU Marine Lab., north Florida, Station M-3.
2/88. x300.
- Fig. 3: *Peridinium divergens* Ehr.
Choctawhatchee Bay, north Florida, Station 31.
7/87. x300.
- Fig. 4: *Peridinium oblongum* (Aurivillius) Cleve.
FSU Marine Lab., north Florida, Station M-4.
2/88. x300.
- Fig. 5: *Peridinium conicum* (Grand) Ostenfeld & Schmidt.
FSU Marine Lab., north Florida, Station M-3.
2/88. x300.
- Figs. 6,7 & 10: *Peridinium oblongum* (Aurivillius) Cleve.
FSU Marine Lab., north Florida, Station M-4.
2/88. x300.
- Figs. 8,9: *Peridinium venustum* Metzenauer.
FSU Marine Lab., north Florida, Station M-4.
2/88. x500.
- Figs. 11,12: *Peridinium oblongum* (Aurivillius) Cleve.
FSU Marine Lab., north Florida, Station M-4.
2/88. x500.
- Fig. 13: *Peridinium divergens* Ehr.
Choctawhatchee Bay, north Florida, Station 31.
7/87. x500.
- Fig. 14: *Peridinium nipponicum* Abe.
Choctawhatchee Bay, north Florida, Station 31.
7/87. x500.
- Fig. 15: *Peridinium divergens* Ehr.
Choctawhatchee Bay, north Florida, Station 31.
7/87. x500.
- Fig. 16: *Peridinium pallidum* Ostenfeld.
Choctawhatchee Bay, north Florida, Station 38.
1/86. x500.

PLATE 6

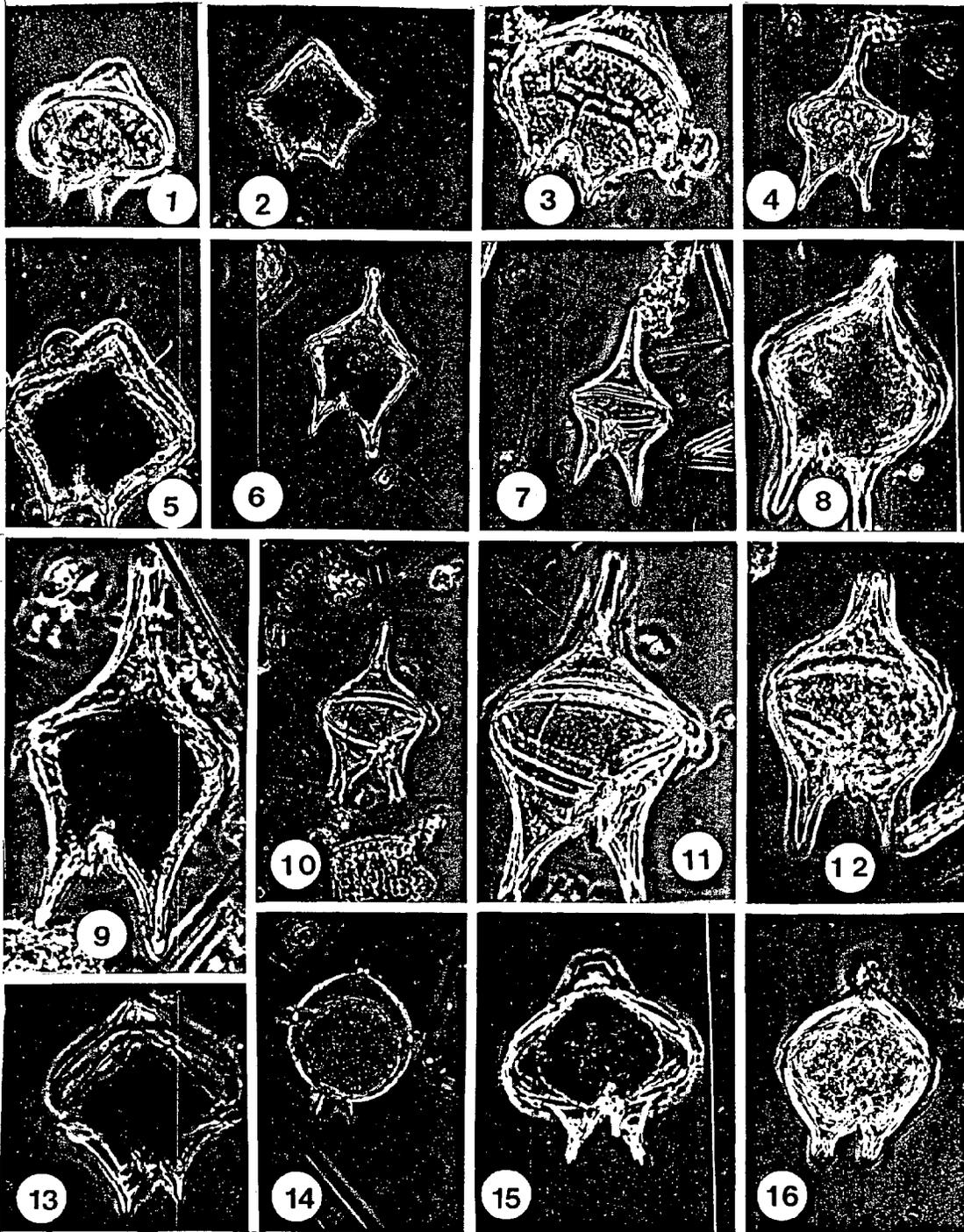


Plate 07

- Fig. 1: *Gymnodinium rotundatum* Klebs.
Choctawhatchee Bay, north Florida, Station 11.
10/87. x1000.
- Figs. 2,4,8: *Prorocentrum micans* Ehr.
Choctawhatchee Bay, north Florida, Station 19.
10/85. x500.
- Fig. 3: *Prorocentrum gracile* Schutt.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x250.
- Fig. 5: *Exuviella baltica* Lohmann
Apalachicola Bay, north Florida, Station E-3.
2/88. x250.
- Fig. 6: *Prorocentrum minimum* Schiller.
Choctawhatchee Bay, north Florida, Station C-3.
10/87. x500.
- Fig. 7: *Dinophysis caudata* Saville-Kent.
Choctawhatchee Bay, north Florida, Station 34.
3/86. x500.
- Fig. 9: *Diplopsalis lenticula* Bergh.
Choctawhatchee Bay, north Florida, Station 31.
12/85. x650.
- Figs. 10, 11: *Diplopsalis lenticula* Bergh.
Apalachicola Bay, north Florida, Station E-3.
2/88. x250.
- Figs. 12, 13: *Ornithoceros magnificus* Stein.
Choctawhatchee Bay, north Florida, Station C-2.
10/87. x200.
- Figs. 14, 17: *Diplopsalis lenticula* Bergh.
Choctawhatchee bay, north Florida, Station 36.
12/86. x600.
- Fig. 15: *Phalacroma cuneus* Schutt.
Choctawhatchee bay, north Florida, Station 15.
10/85. x500.
- Fig. 16: *Cladopyxis brachiolata* Stein
Choctawhatchee bay, north Florida, Station C-2.
10/87. x300.

PLATE 7

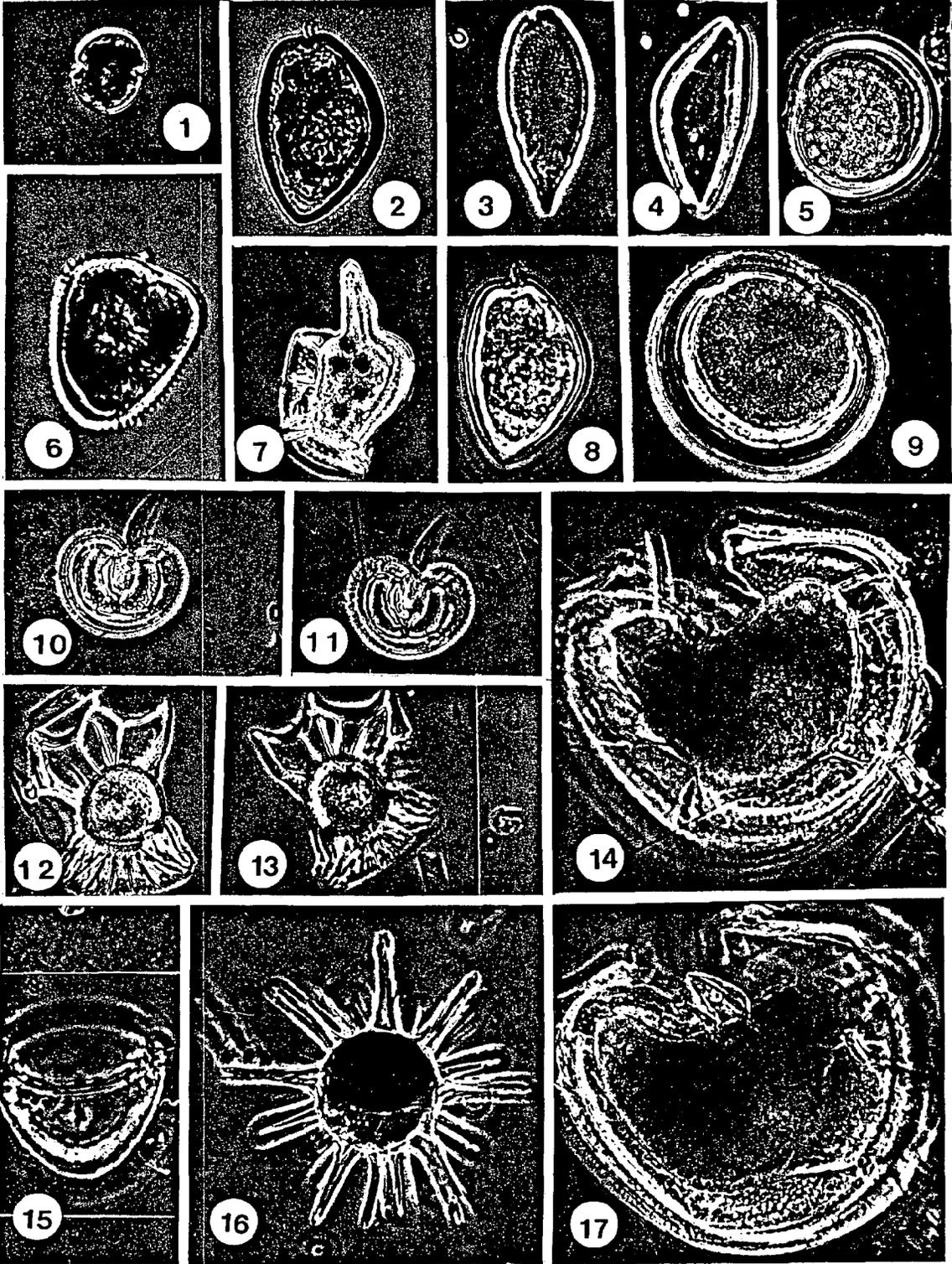


Plate 08

- Fig. 1: *Melosira undulata* (Ehr.) Kuetz.
Choctawhatchee Bay, north Florida, Station 11.
1/86. x500.
- Fig. 2: *Hyalodiscus radiatus* (O'Meara) Grun.
Choctawhatchee Bay, north Florida, Station 11.
1/86. x500.
- Fig. 3: *Podosira stelliger* (Bailey) Mann.
Choctawhatchee Bay, north Florida, Station 03.
1/86. x500.
- Fig. 4: *Stephanopyxis turris* (Grev. & Arn.) Ralfs.
Apalchee Bay, north Florida, Station A-1.
10/87. x200.
- Figs. 5,6: *Stephanopyxis palmeriana* (Grev.) Grun.
Apalachee Bay, north Florida, Station A-3.
10/87. Fig. 5: x250. Fig. 6: x500.
- Fig. 7: *Cyclotella striata* (Kuetz.) Grun.
Choctawhatchee Bay, north Florida, Station 22.
7/86. x1000.
- Fig. 8: *Cyclotella choctawhatcheensis* Prasad sp. nov.
Choctawhatchee Bay, north Florida, Station 19.
4/86. x1200.
- Fig. 9: *Stephanopyxis palmeriana* (Grev.) Grun.
Apalachee Bay, north Florida, Station A-3.
10/87. x800.
- Fig. 10: *Cyclotella meneghiniana* Kuetz.
Choctawhatchee Bay, north Florida, Station 19.
4/86. x1250.
- Figs. 11,12,14: *Cyclotella choctawhatcheensis* Prasad sp. nov.
Choctawhatchee bay, north Florida, Station 15.
4/86. x1200.
- Fig. 13: *Cyclotella cryptica* Reimann et al.,
Choctawhatchee Bay, north Florida, Station 19.
4/86. x1200.
- Figs. 15,16: *Skeletonema costatum* (Grev.) Cleve.
Apalachee Bay, north Florida, Station A-1.
2/88. x500.

PLATE 8

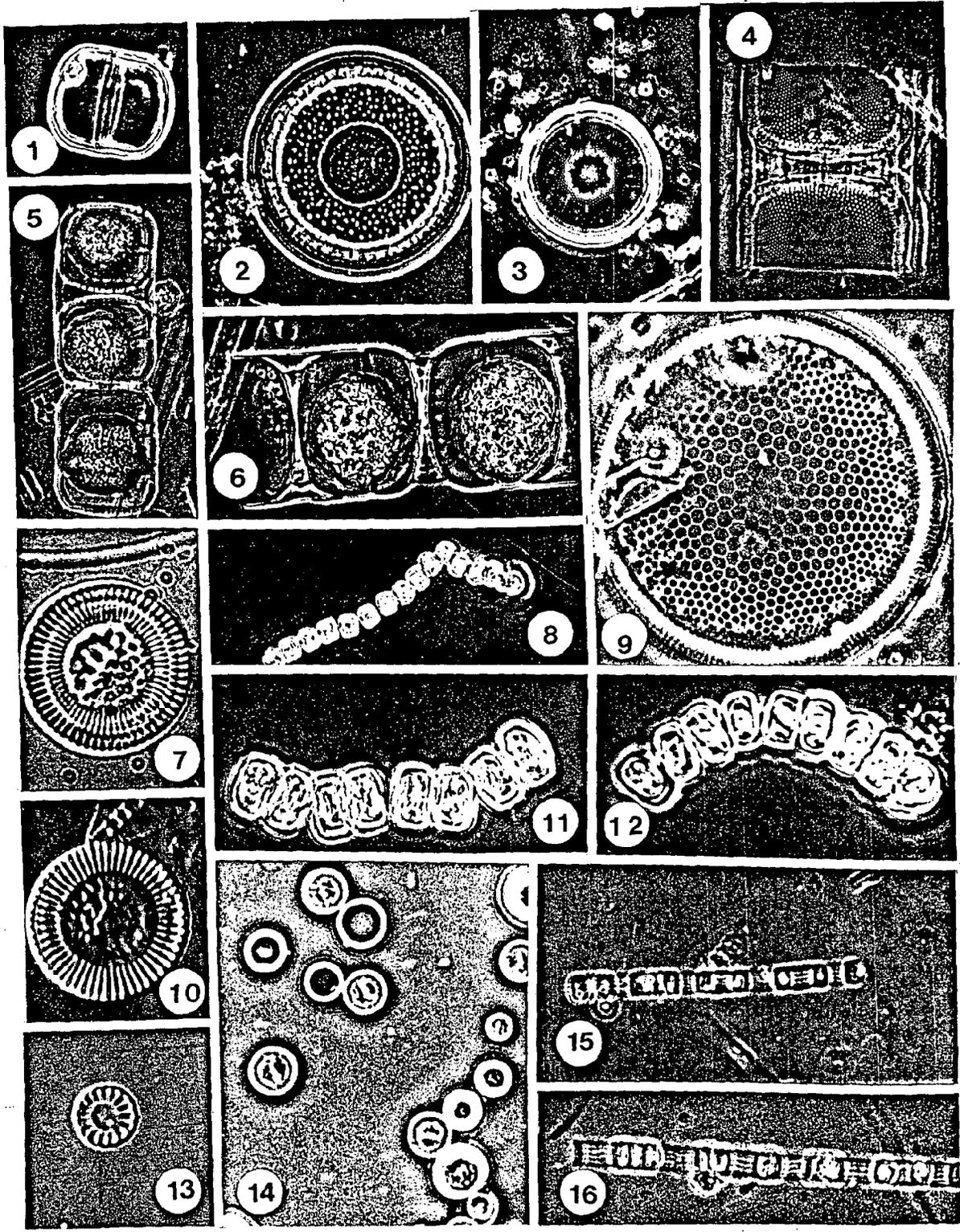


Plate 09

- Figs. 1,2: *Thalassiosira decipiens* (Grun.) Jorg.
Choctawhatchee Bay, north Florida, Station 03.
11/87. x1250.
- Fig. 3: *Thalassiosira oestrupii* (Ostenf.) Pros.-Lev.
Choctawhatchee Bay, north Florida, Station 22.
7/86. x1250.
- Figs. 4,5: *Thalassiosira eccentrica* (Ehr.) Cleve.
Choctawhatchee Bay, north Florida, Station 22.
7/86. x1250.
- Fig. 6: *Thalassiosira tumida* (Jan. ex A.S.) Hasle.
Choctawhatchee Bay, north Florida, Station 22.
7/86. x1250.
- Fig. 7: *Bacteriastrum hyalinum* Lauder.
FSU Marine lab., north Florida, Station 00
2/88. x500.
- Fig. 8: *Lauderia borealis* Gran.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x500.
- Figs. 9,10,11: *Thalassiosira oestrupii* (Ostenf.) Pros.- Lev.
Choctawhatchee bay, north Florida, Station 19.
10/85. x1250.
- Fig. 12: *Thalassiosira lineatus* Jouse.
Choctawhatchee Bay, north Florida, Station 22.
7/86. x1250.

Plate 10

PLATE 9

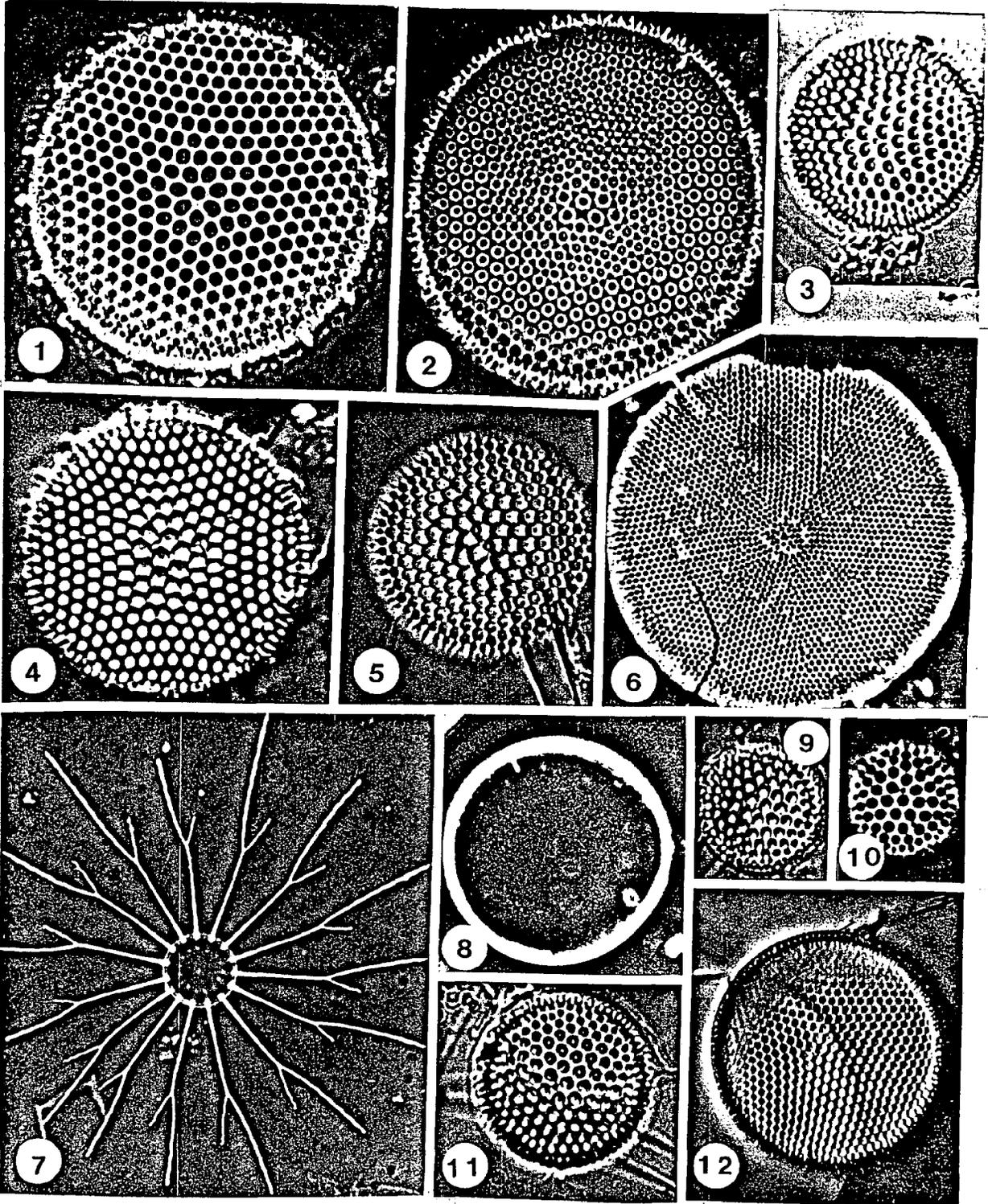


Plate 10

- Fig. 1: *Corethron criophilum* Castracane.
Apalachee Bay, north Florida, Station A-2.
2/88. x500.
- Fig. 2: *Leptocylindrus danicus* Cleve.
Choctawhatchee Bay, north Florida, Station 07.
3/86. x500.
- Figs. 3,4: *Actinoptychus senarius* Ehr.
FSU Marine Lab., north Florida, Station M-4.
2/88. x500.
- Fig. 5: *Azpeitia nodulifer* (A.S.) Fryxell & Sims.
Apalachicola Bay, north Florida, Station E-4.
2/88. x1000.
- Fig. 6: *Actinocyclus ehrenbergii* v. *crassa* (W. Sm.) Hust.
Choctawhatchee Bay, north Florida, Station 03.
1/86. x500.
- Fig. 7: *Corethron criophilum* Castracane.
Apalchee Bay, north Florida, Station A-2.
2/88. x500.
- Fig. 8: *Actinocyclus* sp.
Choctawhatchee Bay, north Florida, Station 03.
1/86. x500.
- Figs. 9,10: *Actinocyclus* cf. *curvatulus* Janisch.
Apalachicola Bay, north Florida, Station E-1.
2/88. x500.
- Fig. 11: *Hemidiscus cuneiformis* Wallich
Apalchee Bay, north Florida, Station A-1.
10/87. x150.
- Fig. 12: *Porosira* cf. *glacialis* (Grun.) Jorg.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x200.
- Fig. 13: *Asteromphallus flabellatus* (Breb.) Grev.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 14: *Actinocyclus ehrenbergii* Ralfs.
Choctawhatchee Bay, north Florida, Station C-3.
10/87. x500.

PLATE 10

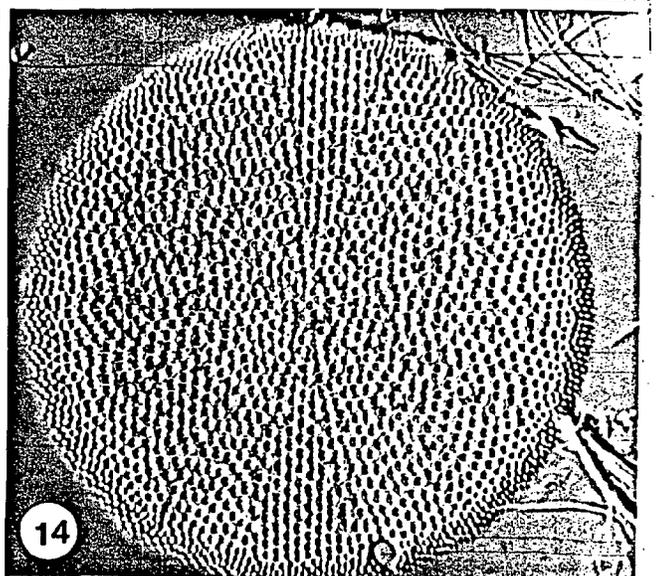
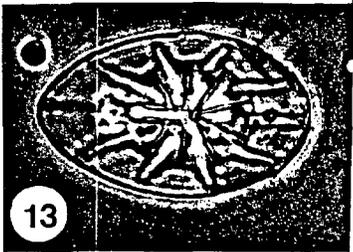
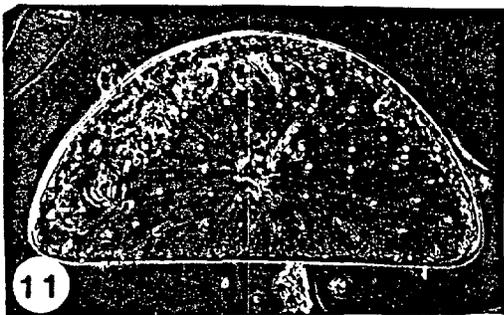
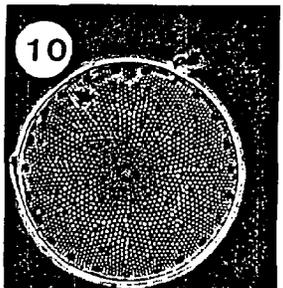
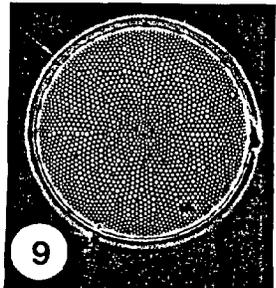
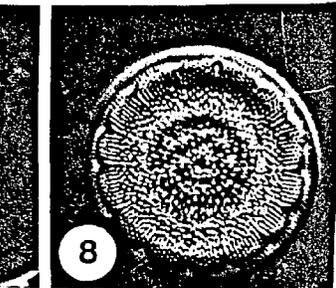
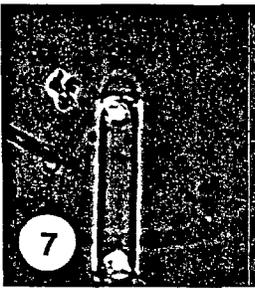
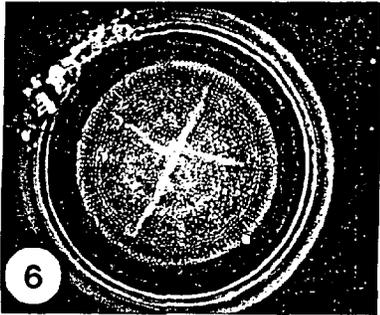
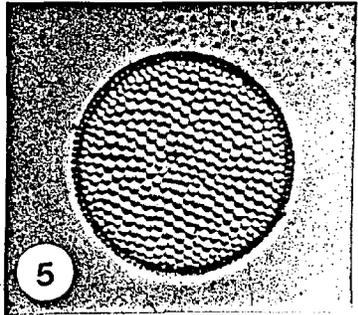
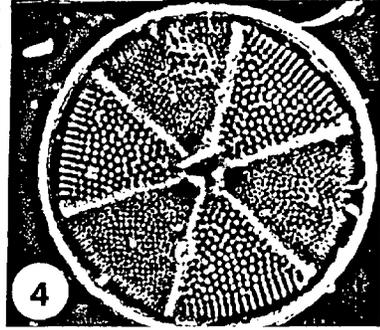
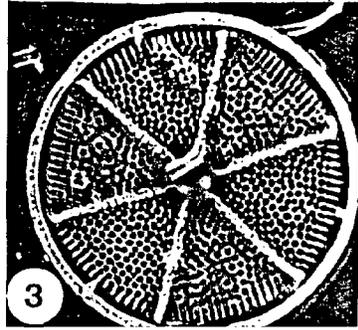
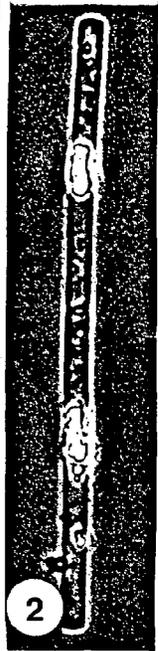
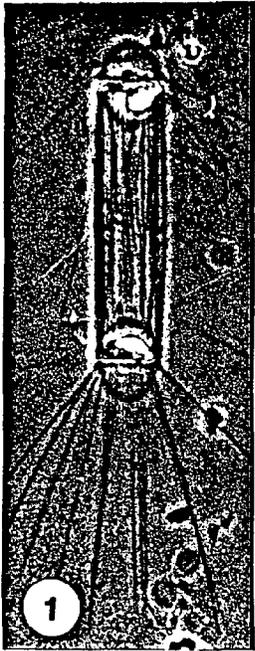


Plate 11

- Figs. 1,2: *Coscinodiscus centralis* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
2/86. x500.
- Fig. 3. *Coscinodiscus apiculatus* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
2/86. x500.
- Fig. 4. *Actinocyclus splendens* (Shadb.) Ralfs.
FSU Marine Lab., north Florida, Station M-4.
2/88. x500.
- Fig. 5: *Coscinodiscus apiculatus* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
2/86. x1200. Central area of the valve in Fig.3.
- Fig. 6: *Coscinodiscus oculus-iridis* Ehr.
FSU Marine Lab., north Florida, Station M-3.
2/87. x1000. Central area of the valve interior.

PLATE 11

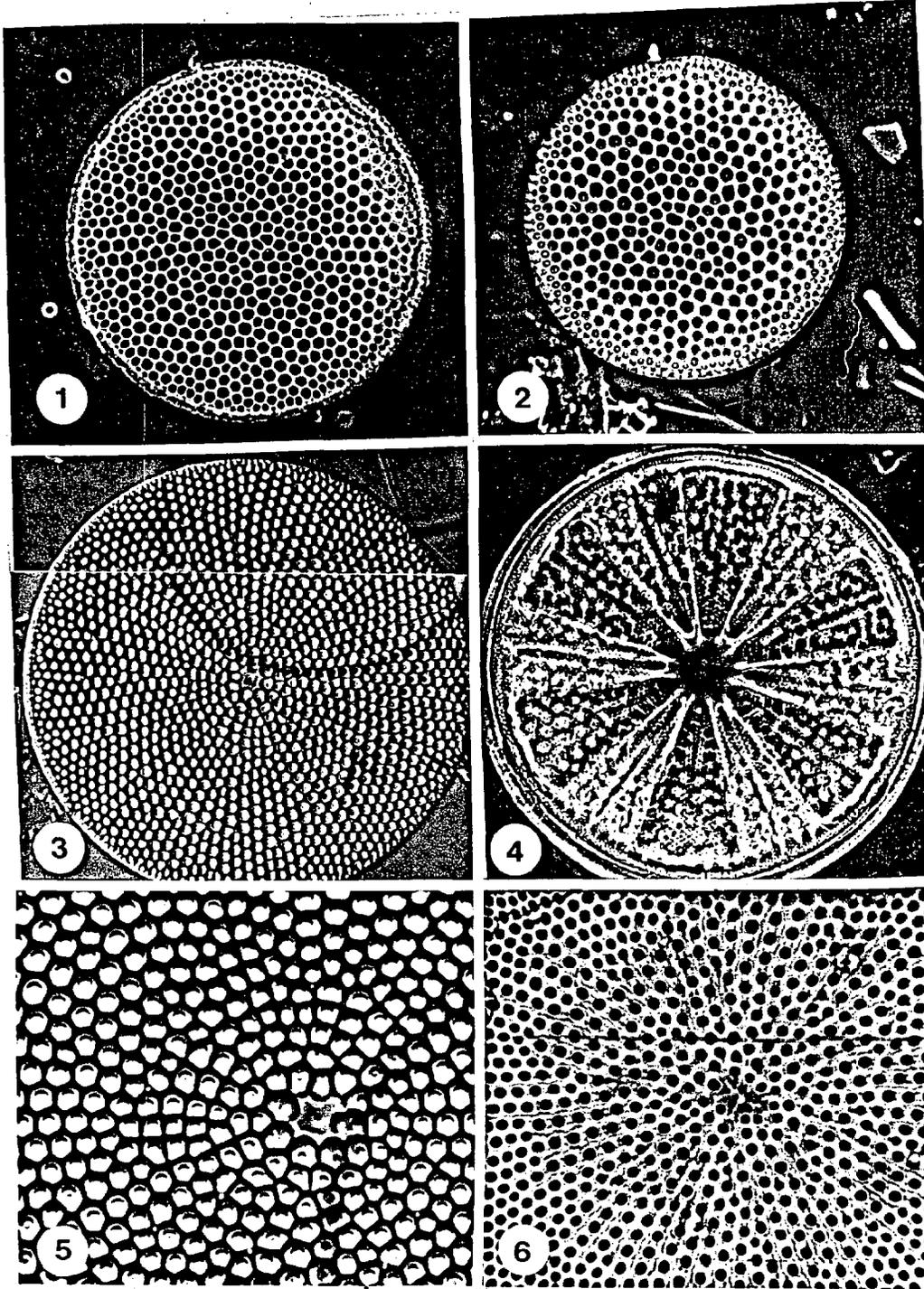


Plate 12

- Figs 1,2: *Coscinodiscus curvatulus* Grun.
Choctawhatchee Bay, north Florida, Station C-3.
2/87. x500.
- Fig. 3: *Melosira undulata* (Ehr.) Kuetz.
Choctawhatchee Bay, north Florida, Station 03.
2/87. x500.
- Fig. 4: *Coscinodiscus radiatus* Ehr.
Choctawhatchee Bay, north Florida, Station 22.
12/85. x500.
- Fig. 5: *Coscinodiscus centralis* Ehr.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.
- Fig. 6: *Eupodiscus radiatus* Bailey
Apalachee Bay, north Florida, Station A-1.
10/87. x250.
- Fig. 7: *Coscinodiscus cf. gigas* Ehr.
Choctawhatchee Bay, north Florida, Station 07.
3/86. x500.
- Fig. 8: *Coscinodiscus centralis* Ehr.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.
- Fig. 9: *Coscinodiscus centralis* Ehr.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.

PLATE 12

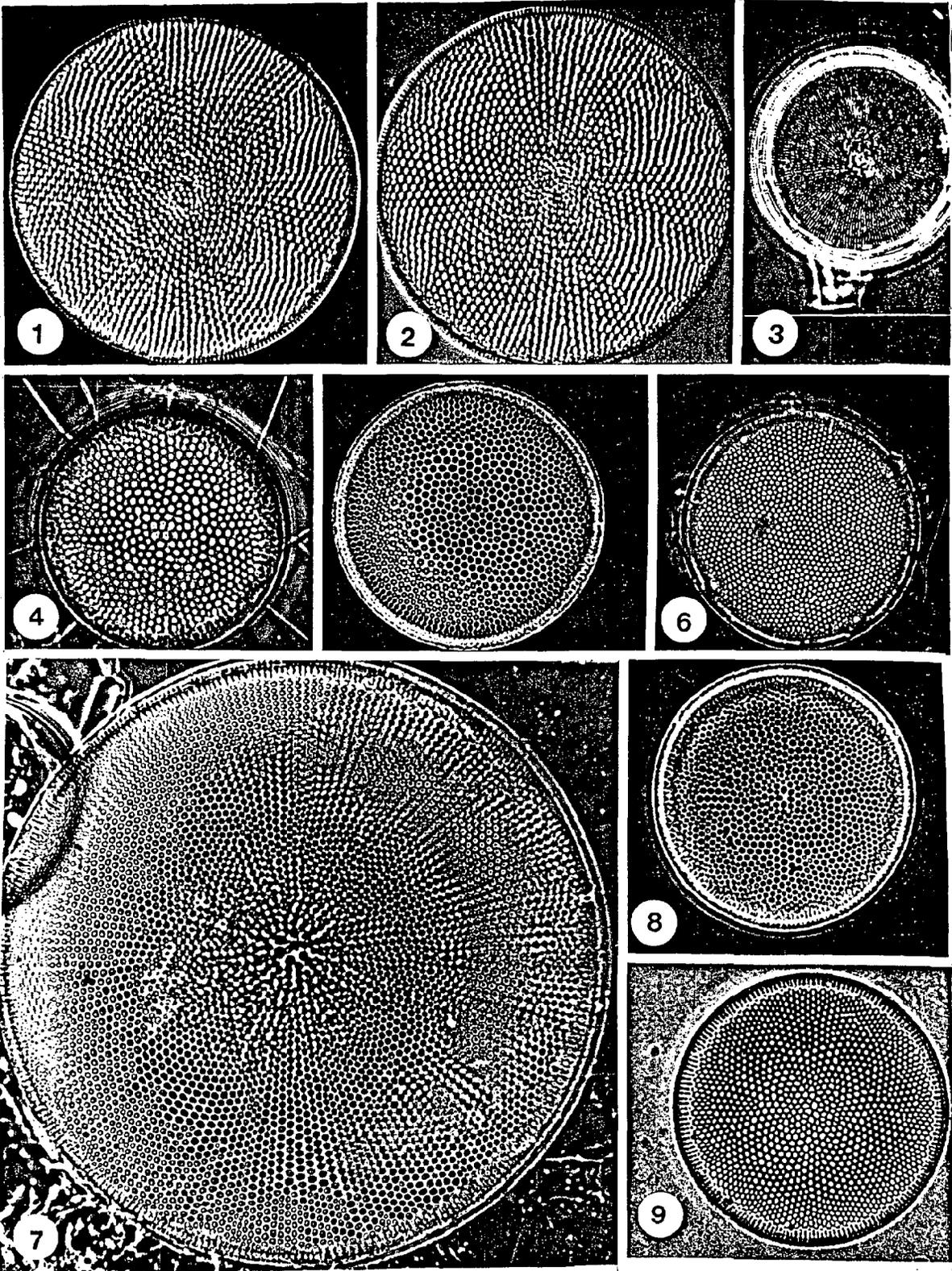


Plate 13

- Fig. 1: *Podosira hormoides* (Mont.) Kuetz.
Choctawhatchee Bay, north Florida, Station 11.
12/85. x500.
- Fig. 2. *Coscinodiscus centralis* Ehr.
Choctawhatchee Bay, north Florida, Station 22.
11/85. x500.
- Fig. 3: *Coscinodiscus oculus-iridis* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 4: *Coscinodiscus oculus-iridis* Ehr.
Choctawhaychee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 5: *Coscinodiscus oculus-iridis* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x1200. Central area enlarged.
- Fig. 6: *Coscinodiscus apiculatus* Ehr.
Choctawhatchee Bay, north Florida, Station 11.
10/85. x500.
- Fig. 7: *Coscinodiscus centralis* Ehr.
Choctawhatchee Bay, north Florida, Station 34.
11/85. x500.
- Fig. 8: *Coscinodiscus oculus-iridis* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x1200.

PLATE 13

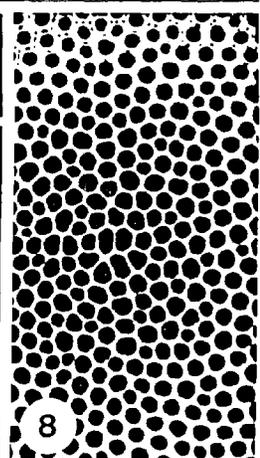
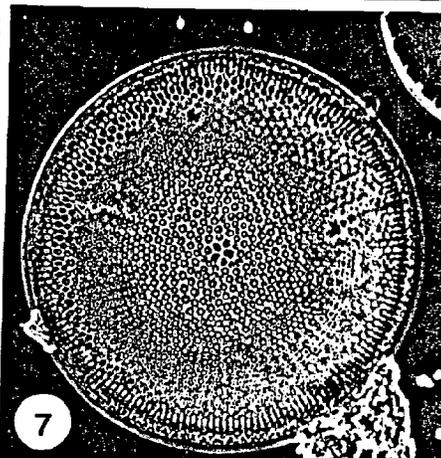
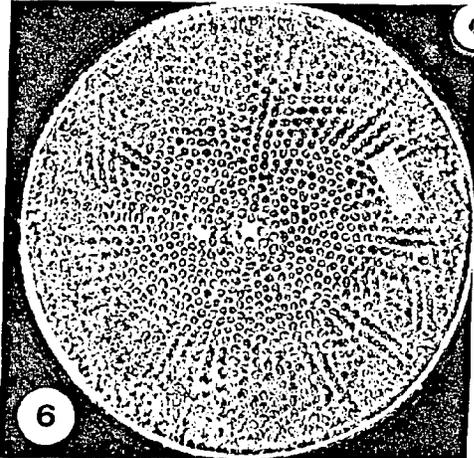
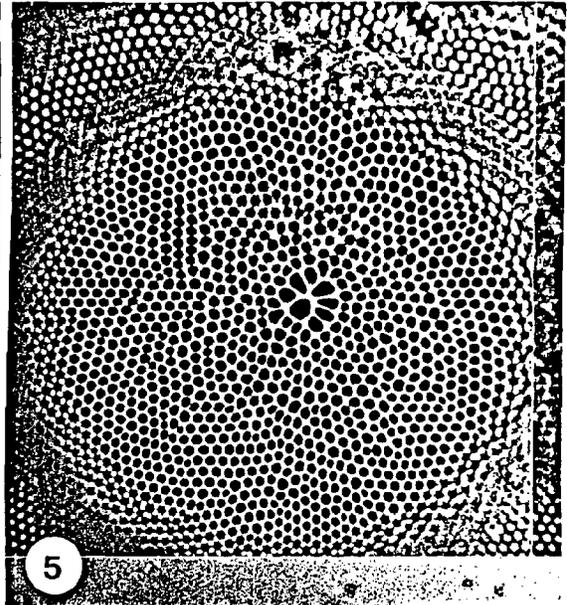
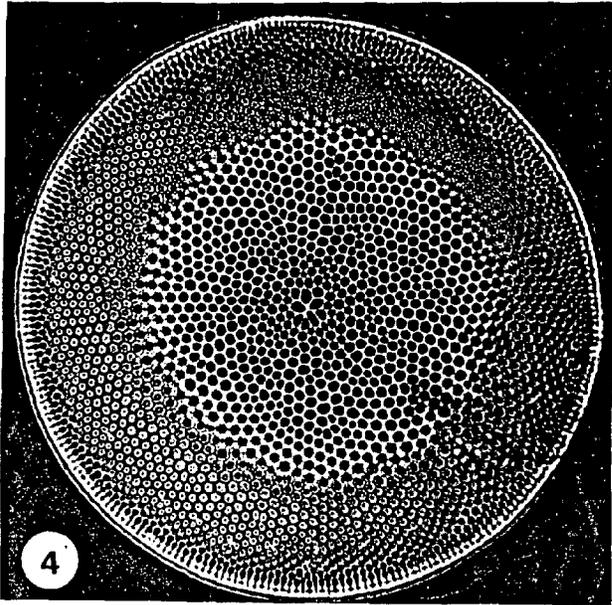
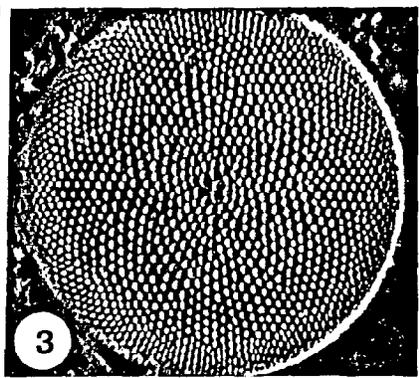
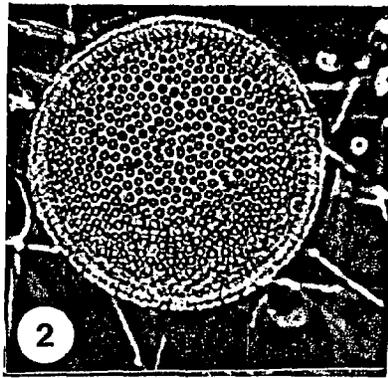
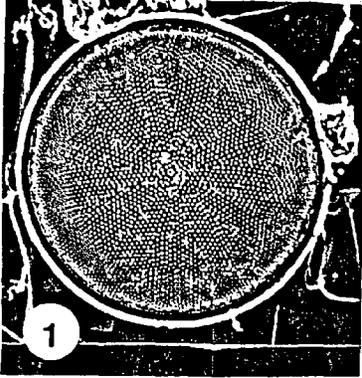


Plate 14

- Fig. 1: *Coscinodiscus curvatulus* Grun.
Choctawhatchee Bay, north Florida, Station 38.
4/86. x500.
- Fig. 2: *Coscinodiscus curvatulus* Grun.
Choctawhatchee Bay, north Florida, Station 38.
4/86. x500.
- Fig. 3: *Coscinodiscus jonesianus* (Grev.) Ostenfeld.
Apalachee Bay, north Florida, Station A-4.
10/87. x250.
- Fig. 4: *Coscinodiscus jonesianus* (Grev.) Ostenfeld.
Apalachee Bay, north Florida, Station A-4.
10/87. x250.
- Fig. 5: *Coscinodiscus radiatus* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
1/86. x1200.
- Fig. 6: *Coscinodiscus jonesianus* (Grev.) Ostenfeld.
Apalachee Bay, north Florida, Station A-4.
10/87. x500.
- Fig. 7: *Coscinodiscus asteromphalus* Ehr.
Choctawhatchee Bay, north Florida, Station 34.
11/85. x750.

PLATE 14

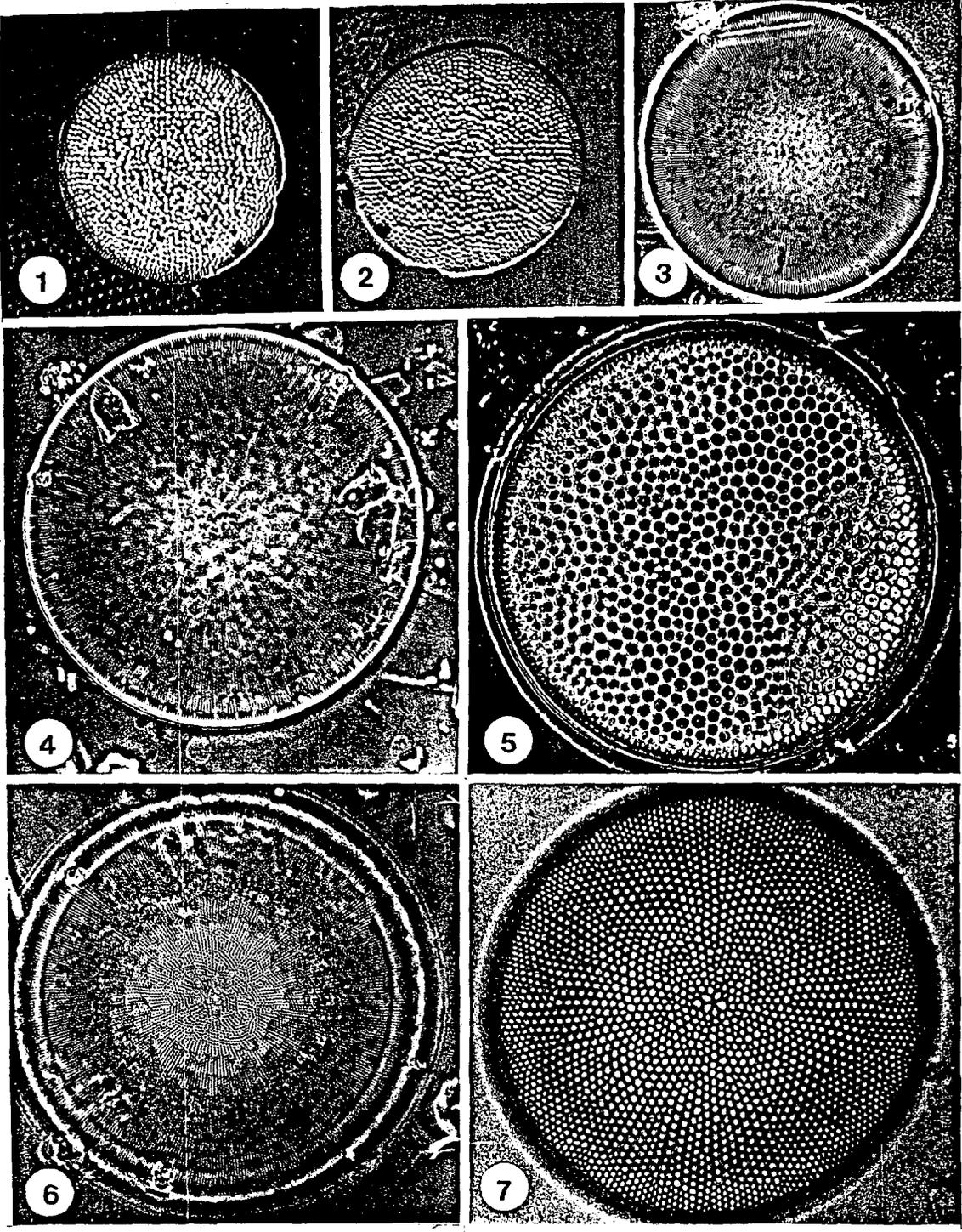


Plate 15

- Fig. 1: *Coscinodiscus perforatus* v. *pavillardii* (Forti) Hustedt. Choctawhatchee bay, north Florida, Station 07. 1/86. x500.
- Fig. 2: *Stellerima microtrias* (Ehr.) Hasle. Apalachee Bay, north Florida, Station A-3. 10/87. x500.
- Fig. 3: *Coscinodiscus granii* Gough. Choctawhatchee Bay, north Florida, station 11. 12/85. x250.
- Fig. 4: *Coscinodiscus granii* Gough. Choctawhatchee Bay, north Florida, Station 25. 10/85. x500.
- Fig. 5: *Coscinodiscus granii* Gough. Choctawhatchee Bay, north Florida, Station 25. 10/85. x500.
- Fig. 6: *Coscinodiscus granii* Gough. Choctawhatchee bay, north Florida, Station 22. 10/85. x1200. A sector enlarged.
- Fig. 7: *Coscinodiscus centralis* Ehr. Choctawhatchee Bay, north Florida, Station 34. 11/85. x500.

PLATE 15

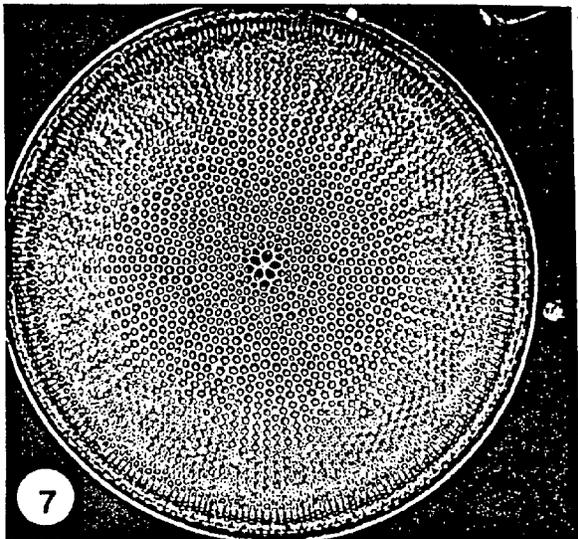
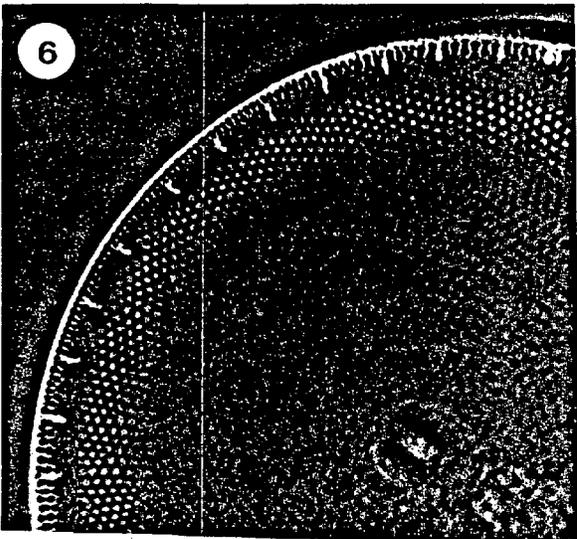
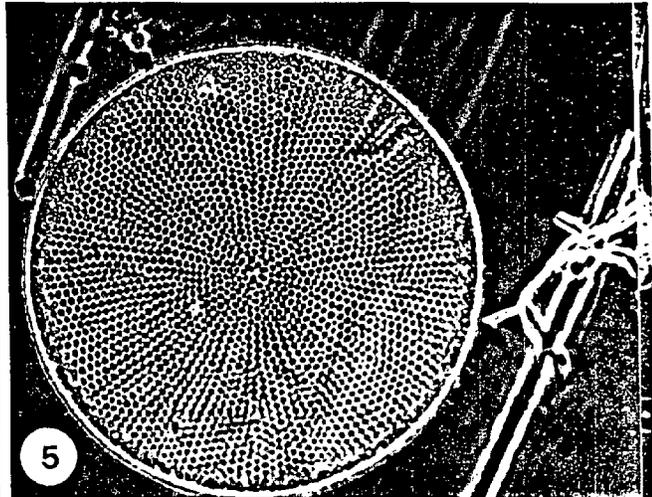
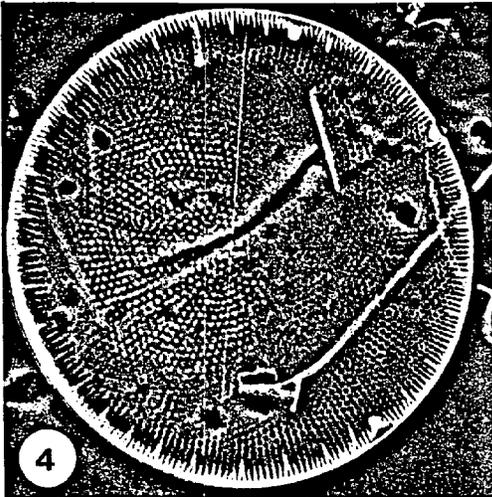
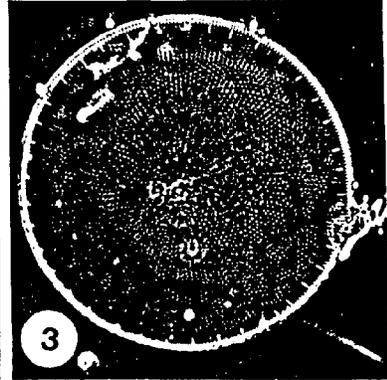
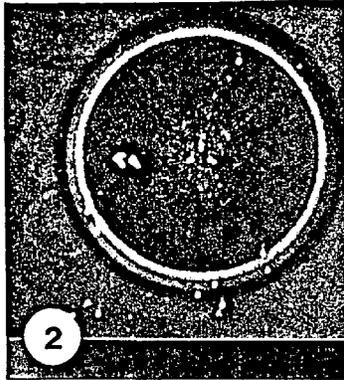
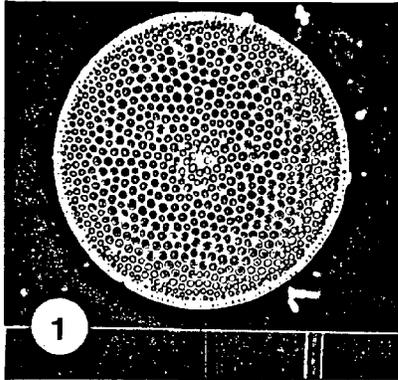


Plate 16

- Figs. 1,3: *Lauderia borealis* Gran.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 2: *Detonula confervacea* (Cleve) Gran.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 4: *Rhizosolenia fragilissima* Bergon.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 5: *Straiatella interrupta* (Ehr.) Heib.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 6: *Rhizosolenia fragilissima* Bergon.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 7: *Rhizosolenia fragilissima* Bergon.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 8: *Guinardia flaccida* (Castr.) Pereg.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 9. *Lauderia borealis* Gran.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 10: *Detonula confervacea* (Cleve) Gran.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Figs.11-13: *Guinardia flaccida* (Castr.) Pereg.
Choctawhatchee Bay, north Florida, Station 22.
11/85. x250.
- Fig. 14: *Rhizosolenia stolterfothii* H. Pereg.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.

PLATE 16

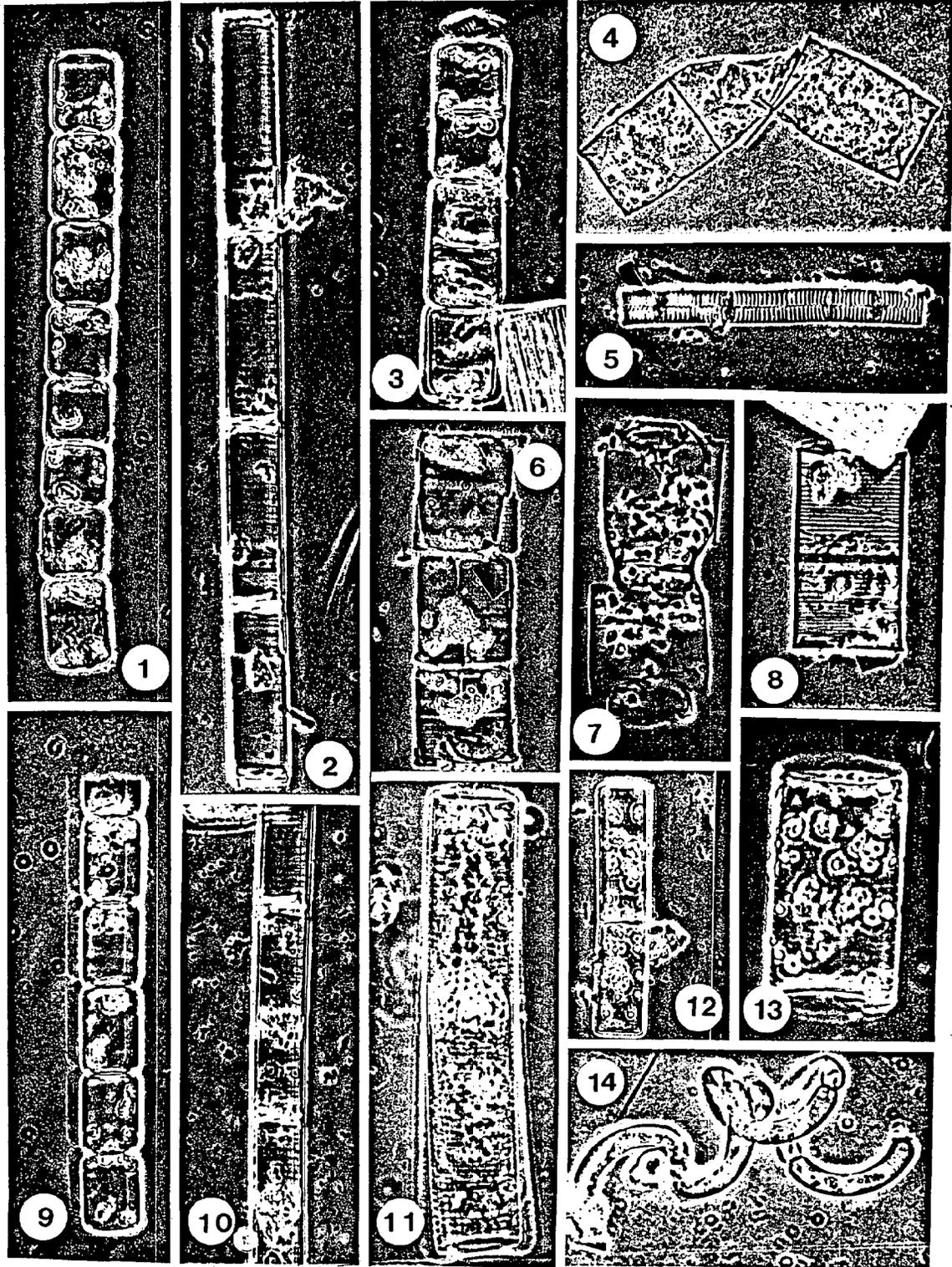


Plate 17

- Fig. 1: *Rhizosolenia robusta* Norman.
Choctawhatchee Bay, north Florida, Station C-02.
10/87. x100.
- Fig. 2: *Rhizosolenia calcar-avis* Schultze.
Apalachee Bay, north Florida, Station A-01.
10/87. x200.
- Fig. 3: *Rhizosolenia alata* Brightwell.
FSU Marine Lab., north Florida, Station M-01.
01/86. x100.
- Fig. 4: *Rhizosolenia castracanei* Peregallo.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x100.
- Figs. 5,6: *Rhizosolenia calcar-avis* Schulze.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Figs. 7,8: *Rhizosolenia robusta* Norman.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x500.
- Fig. 9: *Rhizosolenia imbericata* Brightwell.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x100.
- Fig. 10: *Rhizosolenia stolterfothi* H. Peregallo.
Choctawhatchee Bay, north Florida, Station 38.
3/86. x250.
- Fig. 11: *Rhizosolenia alata* Brightwell.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x100.

PLATE 17

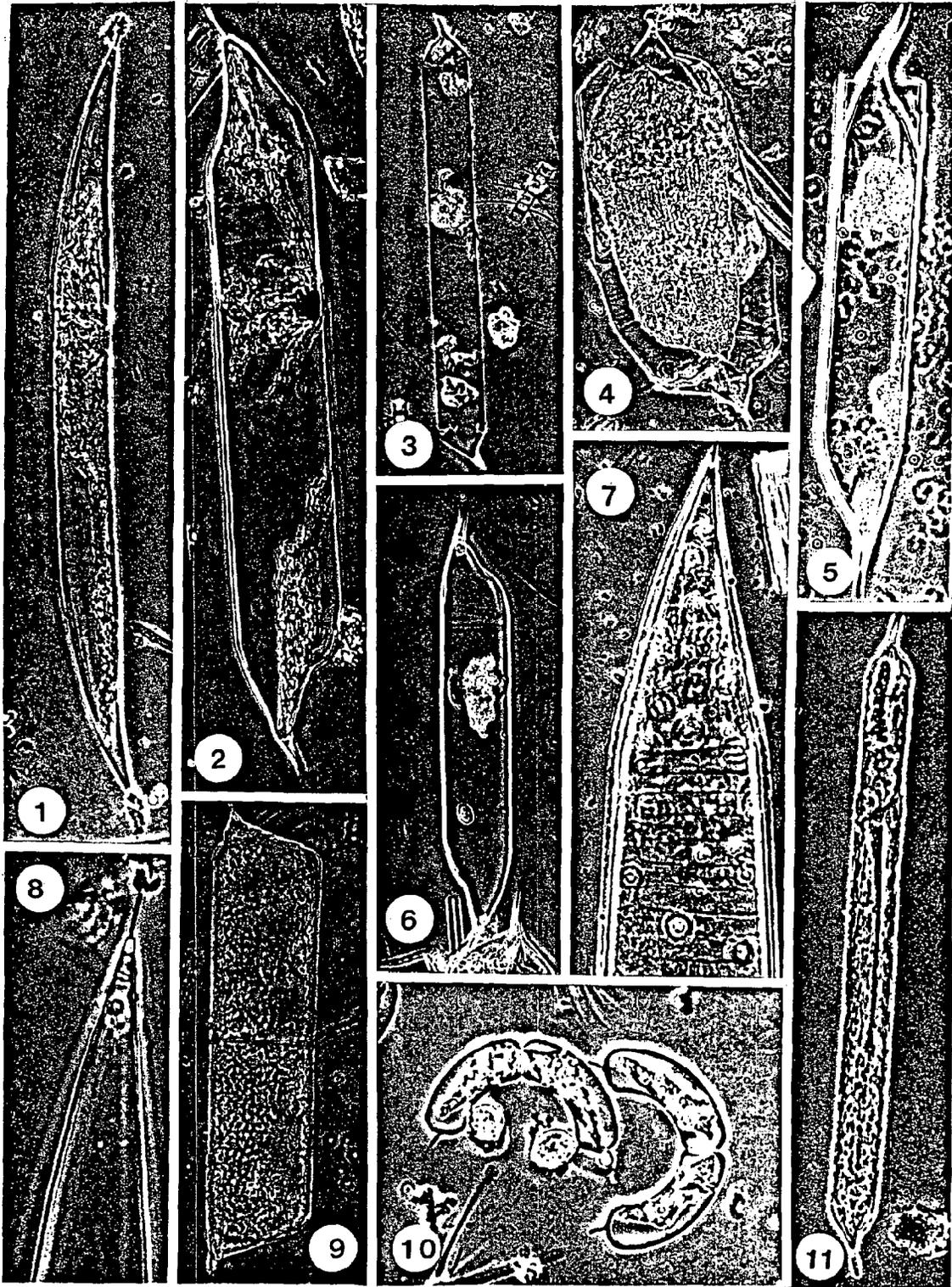


Plate 18

- Fig. 1: *Rhizosolenia fragilissima* Bergon.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x500.
- Fig. 2: *Rhizosolenia setigera* Brightwell.
FSU Marine Lab., north Florida, Station 00.
01/86. x200.
- Fig. 3: *Rhizosolenia alata* Brightwell.
FSU Marine Lab., north Florida, Station 00.
01/86. x500.
- Fig. 4: *Rhizosolenia robusta* Norman.
Apalachee Bay, north Florida, Station A-03.
10/87. x100.
- Fig. 5: *Rhizosolenia robusta* Norman.
Apalachee Bay, north Florida, Station A-03.
10/87. x100.
- Fig. 6: *Rhizosolenia styliformis* Brightwell.
Apalachee Bay, north Florida, Station A-03.
10/87. x100.
- Fig. 7: *Rhizosolenia setigera* Brightwell.
FSU Marine Lab., north Florida, Station 00
1/86. x200.
- Fig. 8: *Rhizosolenia styliformis* Brightwell.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.

PLATE 18

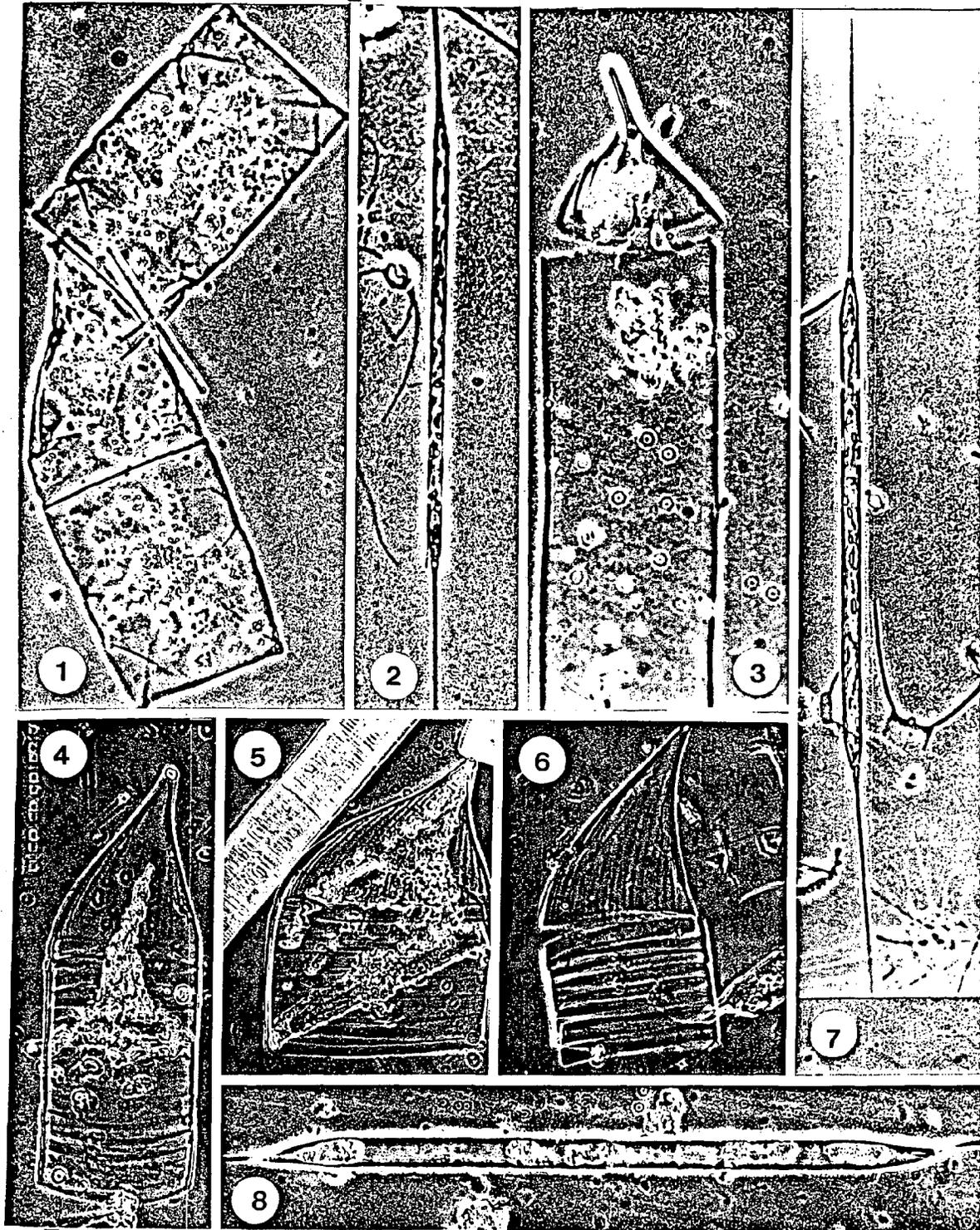


Plate 19.

- Fig. 1: *Rhizosolenia imbricata* Brightwell.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x100.
- Fig. 2: *Rhizosolenia alata* Brightwell.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x100.
- Fig. 3: *Rhizosolenia castracanei* H. Peregallo.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x100.
- Fig. 4: *Rhizosolenia styliiformis* Brightwell.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x500.
- Fig. 5: *Rhizosolenia imbricata* Brightwell.
FSU Marine Lab., north Florida, Station M-04.
2/88. x500.
- Figs. 6,7: *Rhizosolenia imbricata* Brightwell.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. Fig. 6: x100. Fig. 7: x750.
- Figs. 8,9: *Rhizosolenia robusta* Norman.
Choctawhatchee Bay, north Florida, Station C-02.
10/87. Fig. 8: x250. Fig. 9: x100.

PLATE 19

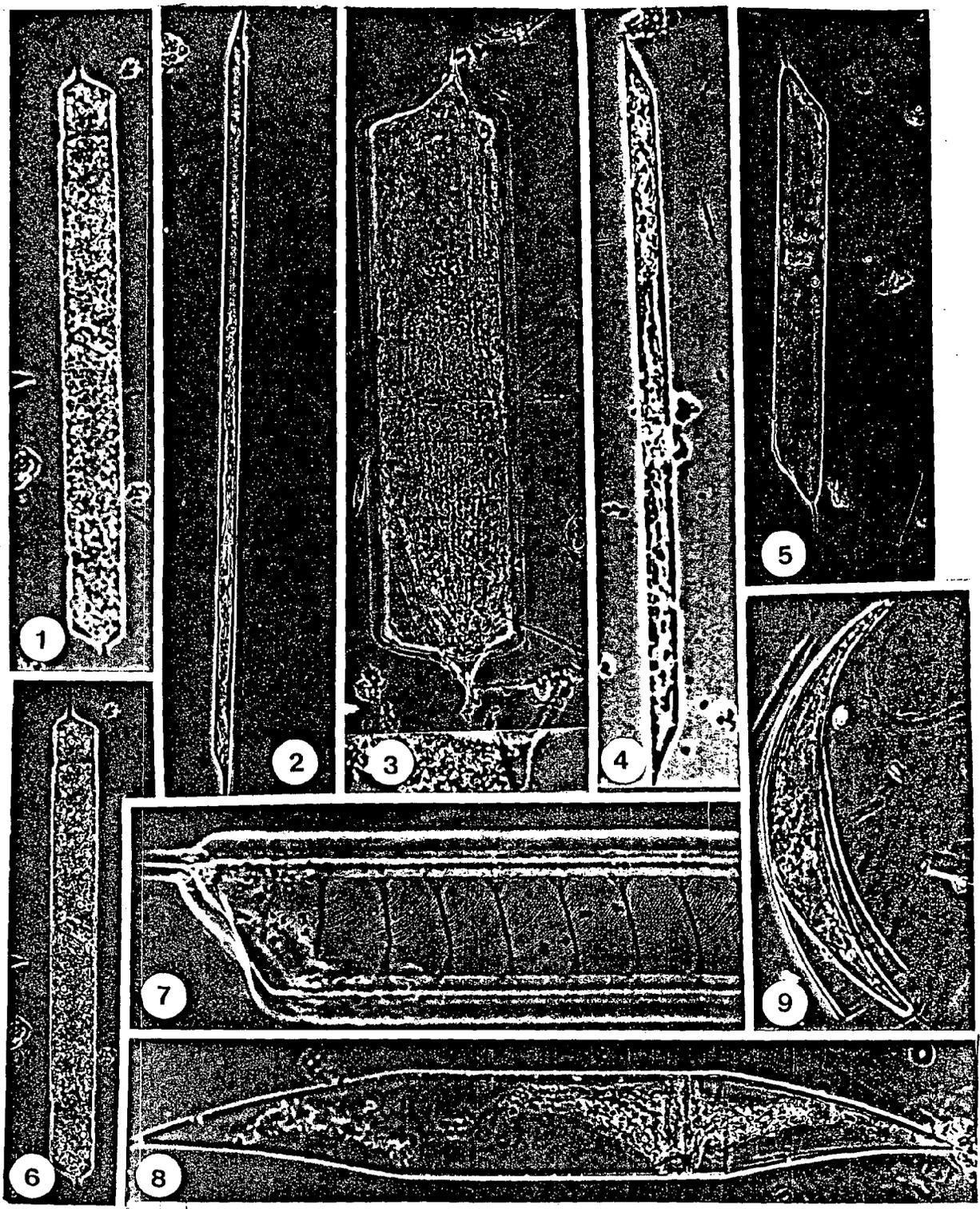


Plate 20

- Fig. 1: *Chaetoceros messanensis* Castrcané.
FSU Marine Lab., north Florida, Station M-03.
02/88. x200.
- Fig. 2: *Chaetoceros diversus* Cleve.
Apalchee Bay, north Florida, Station A-02.
02/88. x200.
- Fig. 3: *Chaetoceros pseudocurvisetus* Mangin.
FSU Marine Lab., north Florida, Station 00.
02/88. x500.
- Fig. 4: *Chaetoceros socialis* Lauder.
Choctawhatchee Bay, north Florida, Station 22.
03/86. x250.
- Fig. 5: *Chaetoceros pseudocrinitum* Ostenfeld.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x500.
- Fig. 6: *Chaetoceros pseudocurvisetus* Mangin.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 7: *Chaetoceros decipiens* Cleve.
Choctawhatchee Bay, north Florida, Station 34.
03/86. x500.
- Fig. 8: *Actinocyclus ehrenbergii* Ralfs.
Choctawhatchee Bay, north Florida, Station 07.
03/86. x250.
- Fig. 9: *Actinocyclus ehrenbergii* v. *tenella* (Breb.) Hust.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.
- Fig. 10: *Coscinodiscus jonesianus* (Grev.) Ostenfeld.
Apalachee Bay, north Florida, Station A-03.
10/87. x200.
- Fig. 11: *Coscinodiscus perforatus* Ehr.
Choctawhatchee Bay, north Florida, Station 03.
01/86. x500.

PLATE 20

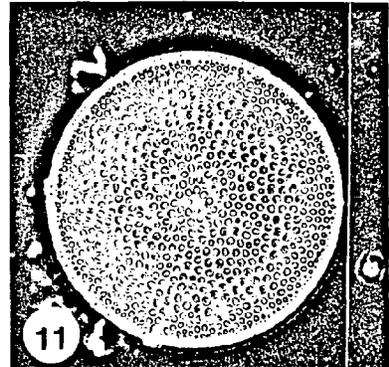
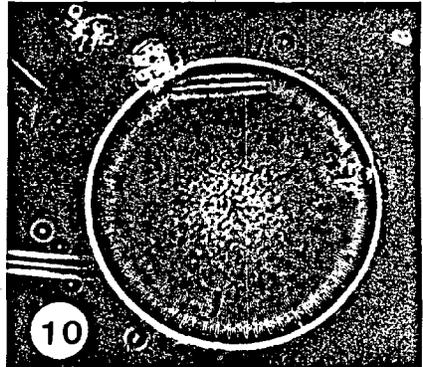
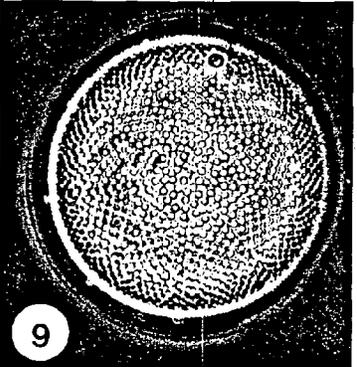
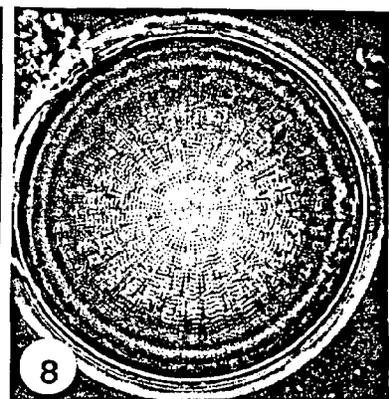
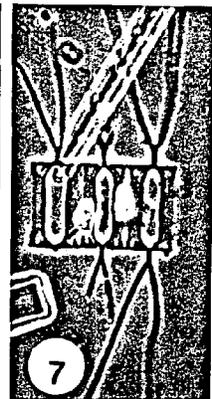
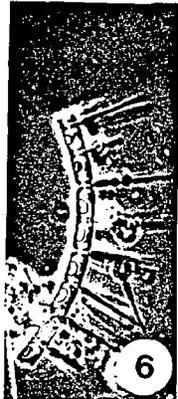
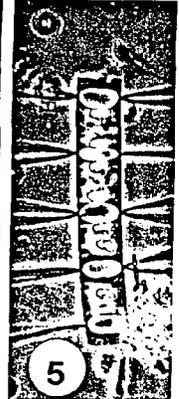
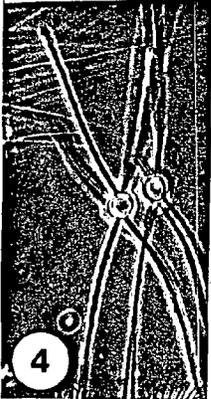
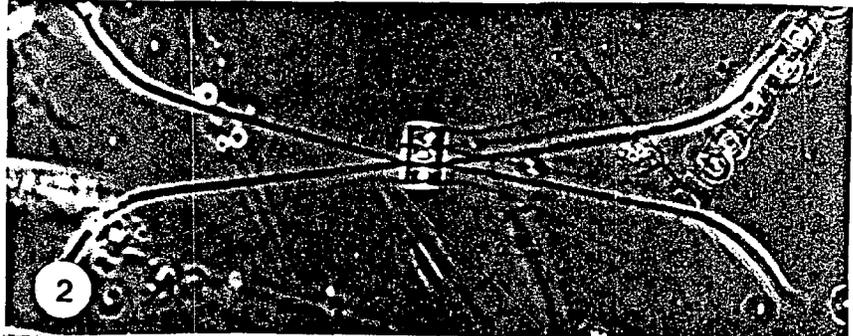
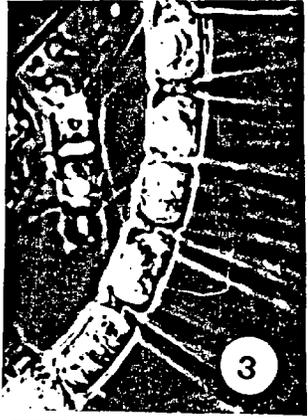
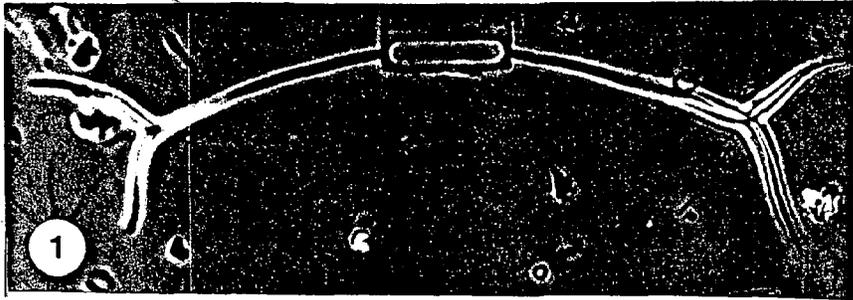


Plate 21

- Figs. 1,2: *Chaetoceros teres* Cleve.
Choctawhatchee bay, north Florida, Station 34.
11/85. x500.
- Fig. 3: *Chaetoceros pseudocurvisetus* Mangin.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x200.
- Fig. 4: *Chaetoceros subsecundes* (Grun.) Hustedt.
Choctawhatchee Bay, north Florida, Station 07.
11/85. x500.
- Fig. 5: *Chaetoceros loranzianus* Grun.
FSU Marine Lab., north Florida, Station 00.
1/86. x500.
- Figs. 6,7: *Bacteriastrum hyalinum* Lauder.
Apalachee Bay, north Florida, Station A-03.
2/88. x500.
- Fig. 8: *Chaetoceros cf. affinis* Lauder.
Choctawhatchee Bay, north Florida, Station 36.
12/85. x500.
- Fig. 9: *Bacteriastrum delicatulum* Cleve.
Choctawhatchee Bay, north Florida, Station 19.
11/85. x500.
- Fig. 10: *Chaetoceros curvisetus* Cleve.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 11: *Chaetoceros decipiens* Cleve.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x500.
- Fig. 12: *Chaetoceros compressus* Lauder
Choctawhatchee Bay, north Florida, Station 35.
10/86. x500.

PLATE 21

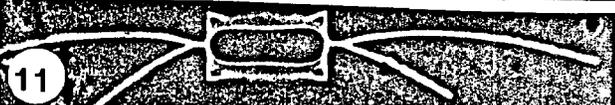
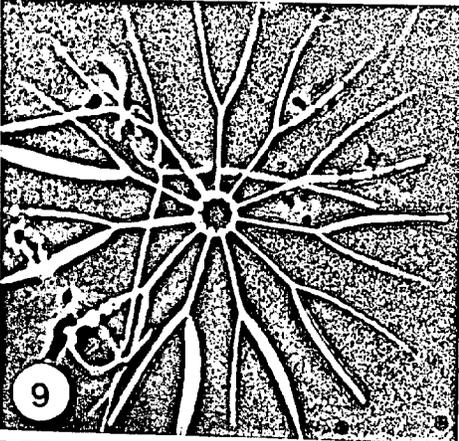
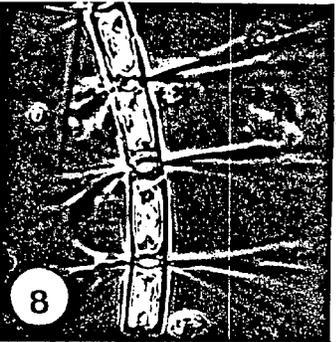
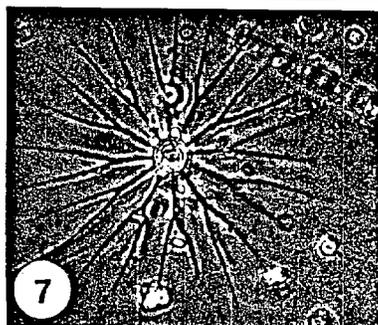
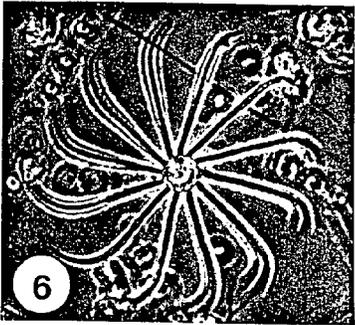
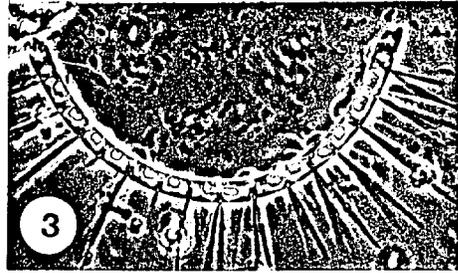
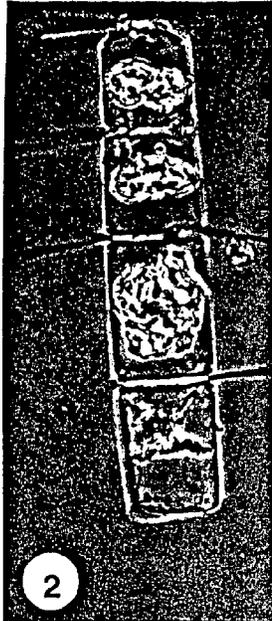
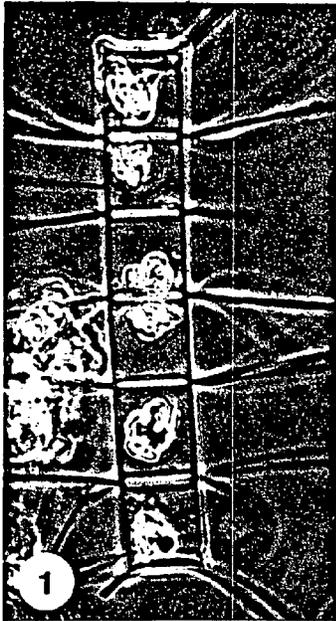


Plate 22

- Fig. 1: *Chaetoceros borealis* Bailey.
Choctawhatchee bay, north Florida, Station 07.
11/85. x500.
- Fig. 2: *Chaetoceros decipiens* Cleve.
Choctawhatchee Bay, north Florida, Station 07.
11/85. x250.
- Fig. 3: *Chaetoceros pseudocurvisetus* Mangin.
FSU Marine Labo., north Florida, Station 00.
01/86. x400.
- Fig. 4: *Chaetoceros pseudocrinitus* Ostenfeld.
Choctawhatchee Bay, north Florida, Station 19.
12/85. x400.
- Fig. 5: *Chaetoceros loranianus* Grun.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x400.
- Fig. 6: *Chaetoceros decipiens* Cleve.
Choctawhatchee bay, north Florida, Station 35.
10/86. x400.
- Fig. 7: *Chaetoceros subsecundus* (Grun.) Hustedt.
Choctawhatchee bay, north Florida, Station 34.
12/85. x400.
- Fig. 8: *Chaetoceros brevis* Schutt.
Choctawhatchee bay, north Florida, Station 35.
01/86. x250.
- Fig. 9: *Chaetoceros cf. affinis* lauder.
Choctawhatchee Bay, north Florida, Station 03.
11/85. x400.

PLATE 22

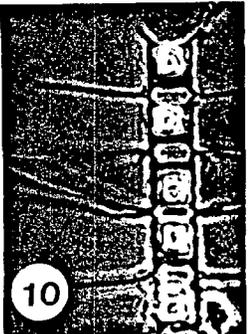
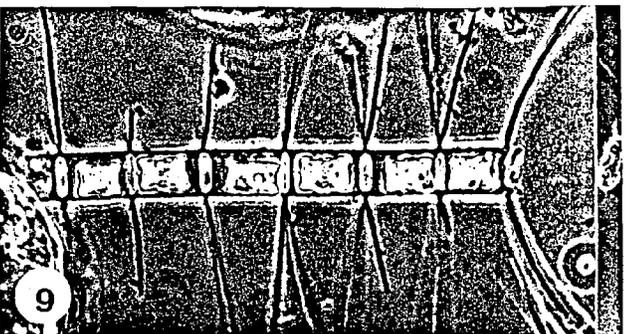
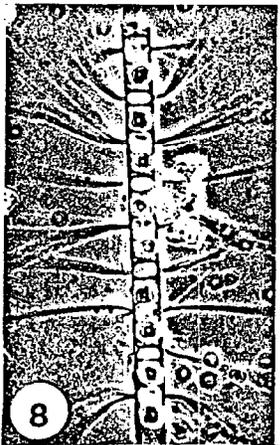
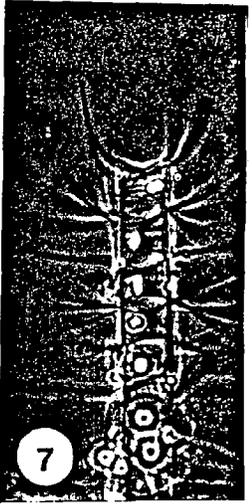
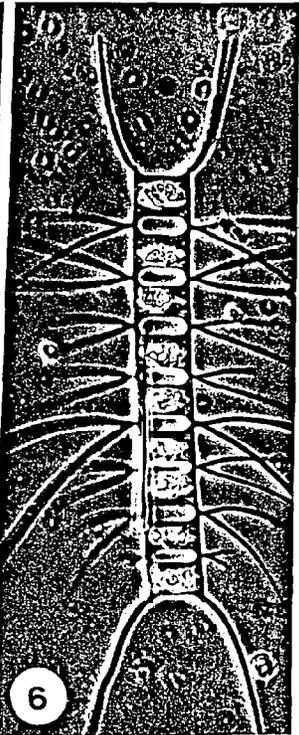
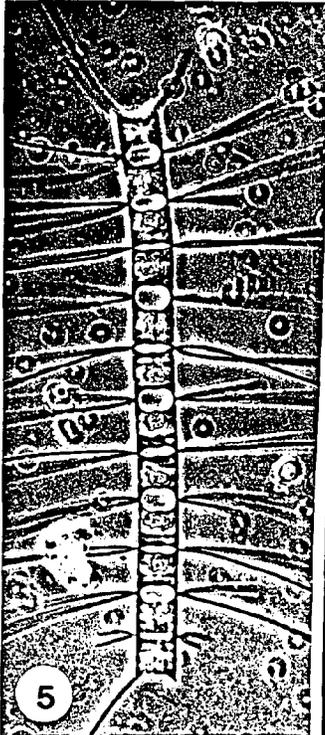
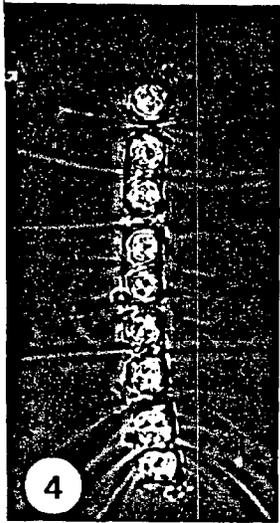
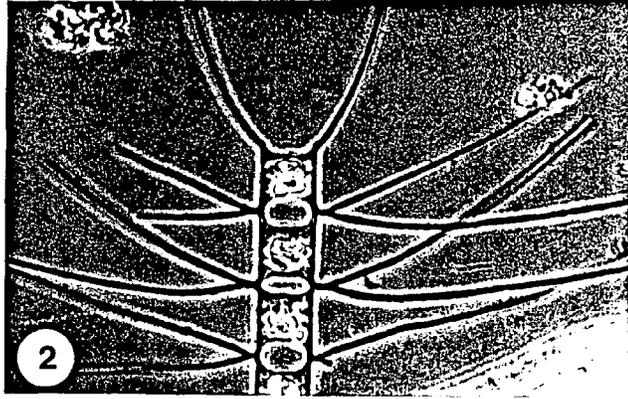
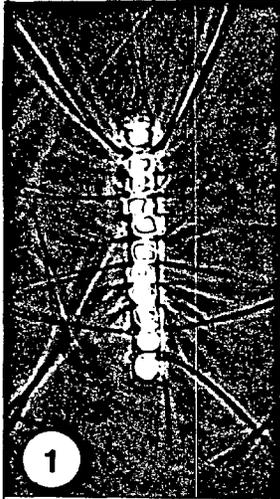


Plate 23

- Fig. 1: *Chaetoceros didymus* Ehr.
Choctawhatchee Bay, north Florida, Station 15.
10/85. x400.
- Fig. 2: *Chaetoceros dichchaeta* Ehr.
Choctawhatchee Bay, north Florida, Station 36.
12/85. x400.
- Fig. 3: *Chaetoceros didymus* v. *protuberance* (Laud.) Gran.
Choctawhatchee Bay, north Florida, Station 34.
03/86. x400.
- Fig. 4: *Chaetoceros dichchaeta* Ehr.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x400.
- Fig. 5: *Chaetoceros peruvianus* Brightwell.
FSU Marine Lab., north Florida, Station 00.
01/86. x500.
- Fig. 6: *Chaetoceros compressus* Lauder.
Choctawhaychee Bay, north Florida, Station 35.
10/86. x400.
- Fig. 7: *Chaetoceros decipiens* Cleve.
Apalachicola Bay, north Florida, Station E-02.
2/88. x200.
- Fig. 8: *Chaetoceros didymus* v. *protuberance* (Laud.) Gran.
Choctawhatchee Bay, north Florida, Station 22.
03/86. x400.
- Fig. 9: *Chaetoceros decipiens* Cleve.
Choctawhatchee Bay, north Florida, Station 07.
11/85. x500.

PLATE 23

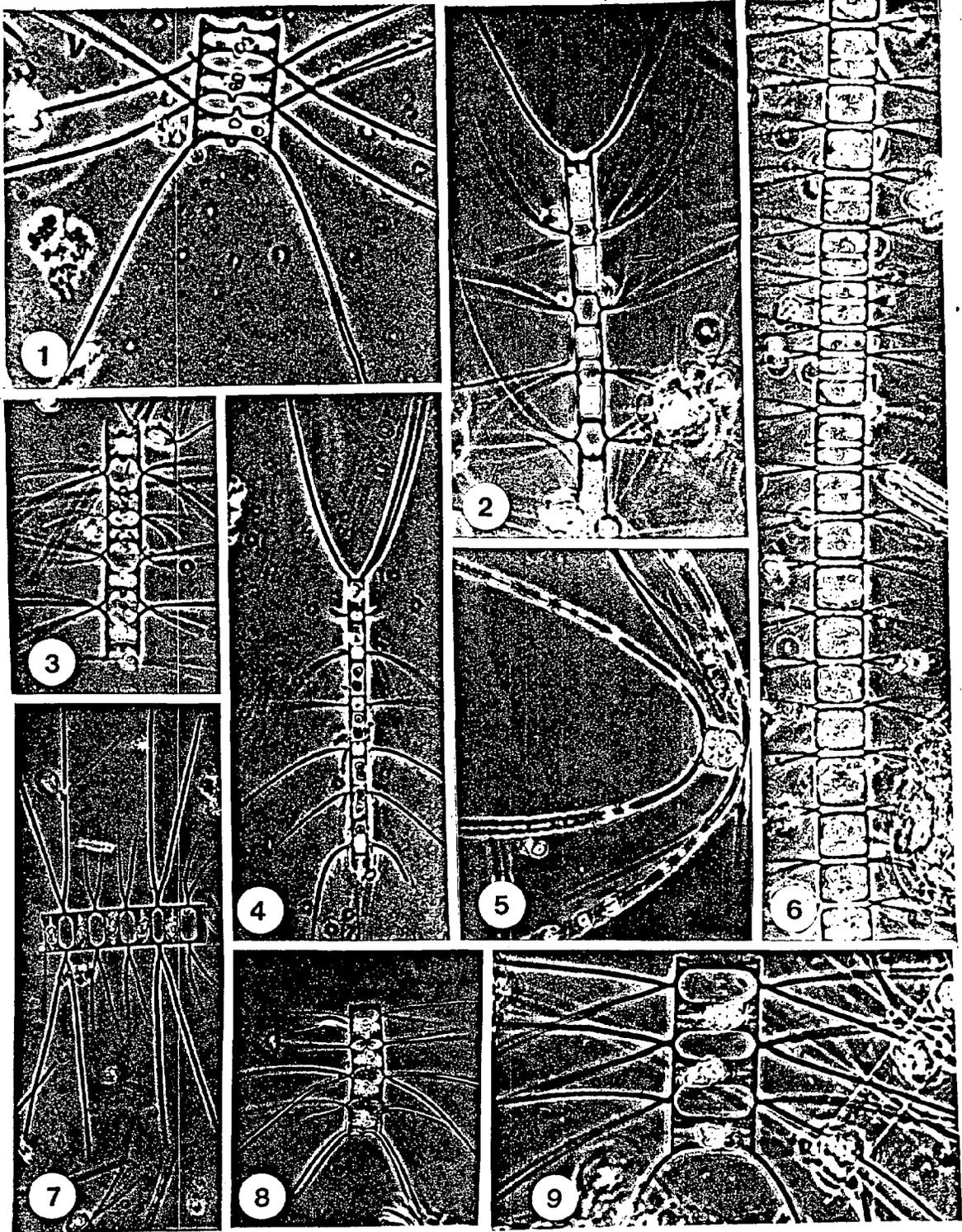


Plate 24

- Fig. 1: *Chaetoceros teres* Cleve
Choctawhatchee bay, north Florida, Station 34.
12/85. x400.
- Fig. 2: *Chaetoceros teres* Cleve.
Choctawhatchee Bay, north Florida, Station 03.
11/87. x400.
- Fig. 3: *Chaetoceros affinis* Lauder.
Choctawhatchee Bay, north Florida, Station 34.
03/86. x400.
- Fig. 4: *Chaetoceros affinis* Lauder.
Choctawhatchee bay, north Florida, Station 22.
03/86. x400.
- Fig. 5: *Chaetoceros eibenii* Grunow.
Choctawhatchee Bay, north Florida, Station 36.
10/85. x400.
- Fig. 6: *Chaetoceros affinis* Lauder.
Choctawhatchee Bay, north Florida, Station 07.
11/85. x400.
- Fig. 7: *Chaetoceros affinis* Lauder.
Choctawhatchee Bay, north Florida, Station 34.
03/96. x400.
- Fig. 8: *Chaetoceros compressus* Lauder.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.

PLATE 24

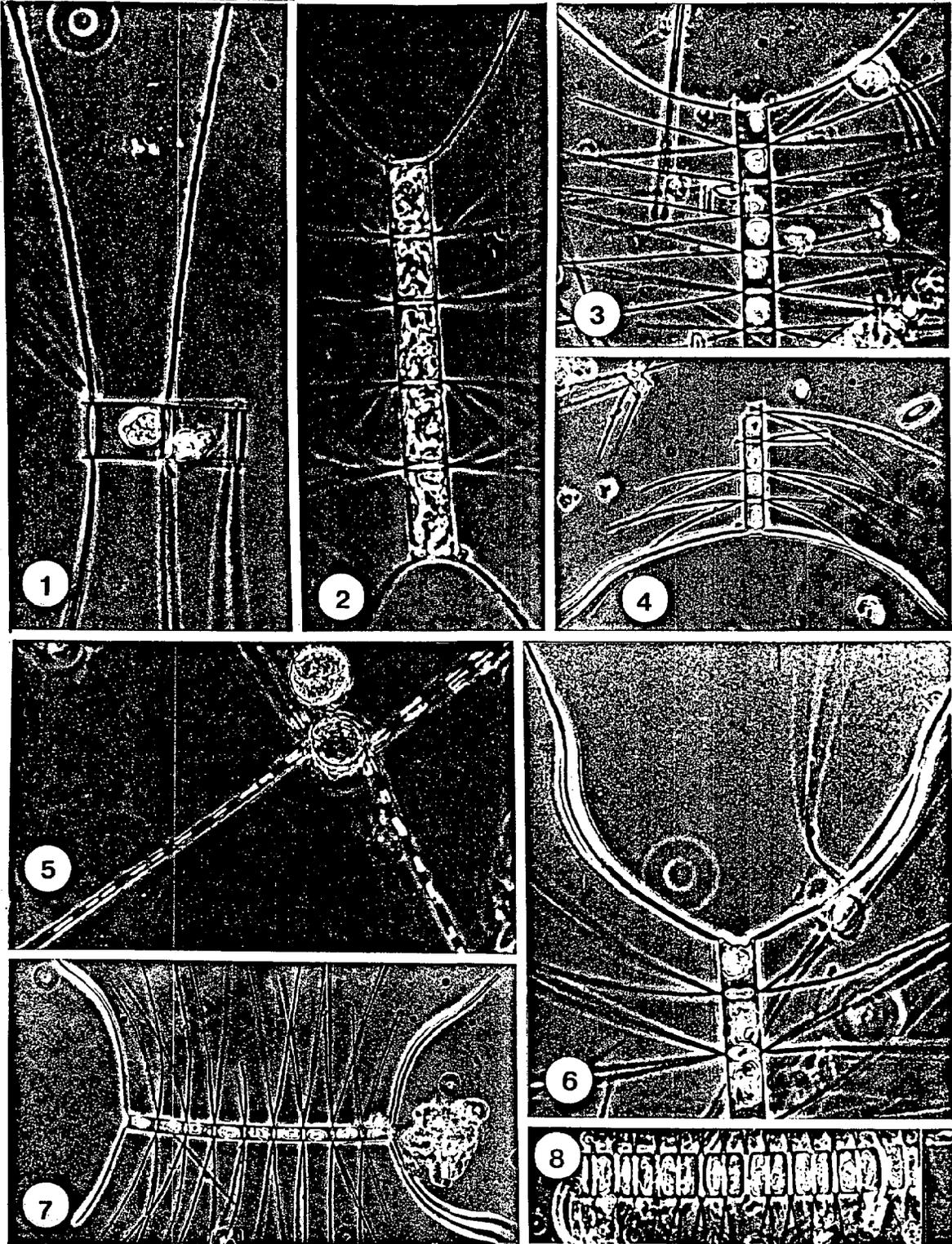


Plate 25

- Figs. 1,2: *Climacodium frauenfeldianum* Grunow.
Choctawhatchee Bay, north Florida, Station C-02.
10/87. Fig. 1: x100. Fig. 2: x200.
- Fig. 3: *Hemiaulus sinensis* Greville.
Apalachee Bay, north Florida, Station A-04.
10/87. x200.
- Fig. 4: *Hemiaulus haukii* Grunow.
Choctawhatchee Bay, north Florida, Station 22.
03/86. x400.
- Fig. 5: *Hemiaulus membranaceus* Cleve.
FSU Marine Lab., north Florida, Station M-03.
2/88. x250.
- Fig. 6: *Hemiaulus sinensis* Greville.
Apalachee Bay, north Florida, Station A-4.
10/87. x500.
- Fig. 7: *Hemiaulus membranaceus* Cleve.
Choctawhatchee bay, north Florida, Station C-03.
10/87. x250.
- Fig. 8: *Hemiaulus sinensis* Greville.
Choctawhatchee Bay, north Florida, station 35.
10/86. x200.
- Fig. 9: *Hemiaulus membranaceus* Cleve.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 10: *Attheya decora* West.
Choctawhatchee Bay, north Florida, Station 34.
03/86. x500.
- Fig. 11: *Hemiaulus membranaceus* Cleve.
Apalachee Bay, north Florida, Station A--02.
02/88. x500.

PLATE 25

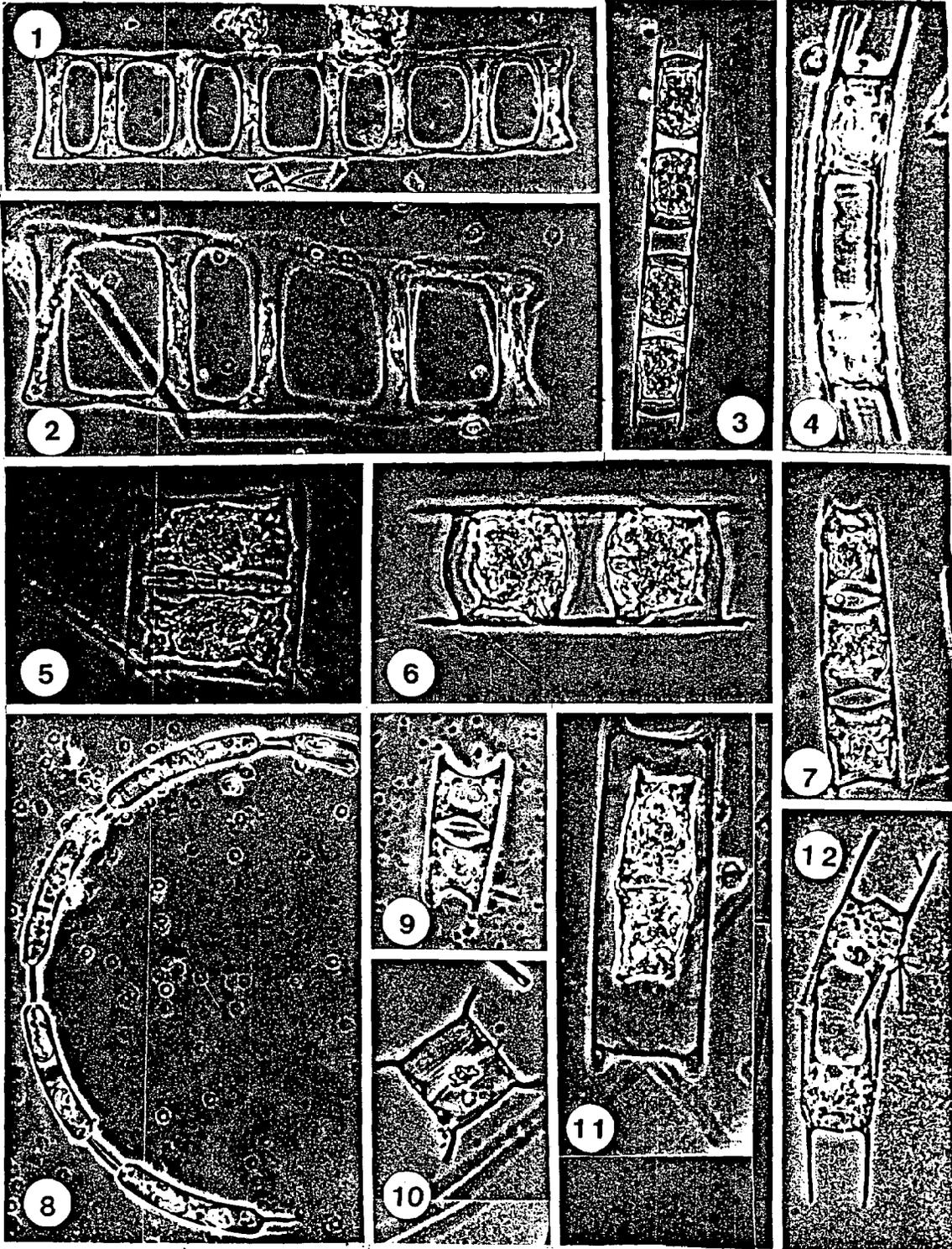


Plate 26

- Figs. 1,2: *Ditylum brightwellii* (West) Grunow.
Apalachee Bay, north Florida Station A-03.
02/88. Fig. 1: x500. Fig. 2: x250.
- Fig. 3: *Biddulphia sinensis* Greville.
Choctawhatchee Bay, north Florida, Station 38.
12/85. x250.
- Fig. 4: *Cymatosira lorenziana* Grunow.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x250.
- Fig. 5: *Biddulphia mobiliensis* Bailey.
Apalachee Bay, north Florida, Station A-01.
02/88. x500.
- Figs. 6,7: *Cymatosira lorenziana* Grunow.
Choctawhatchee Bay, north Florida, Station 36.
12/85. x1000.
- Fig. 8: *Biddulphia sinensis* Greville.
Apalachee Bay, north Florida, Station A-03.
10/87. x250.
- Fig. 9: *Biddulphia sinensis* Freville.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 10: *Biddulphia sinensis* Greville.
Choctawhatchee Bay, north Florida, Station 36.
08/87. x250.
- Figs. 11,12: *Eunatogramma laeve* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 13: *Cymatosira belgica* Grunow.
Choctawhatchee Bay, north Florida, Station C-02.
10/87. x250.
- Fig. 14: *Trigonium alternans* (Bailey) Mann.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.

PLATE 26

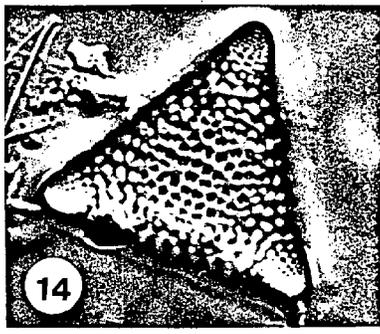
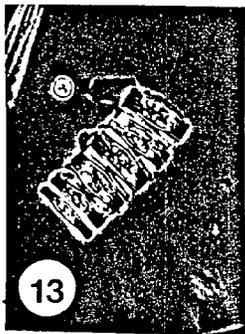
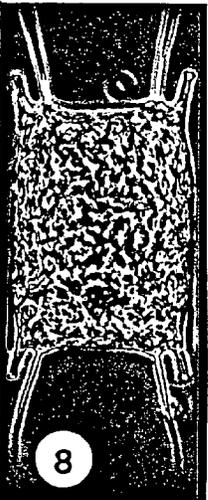
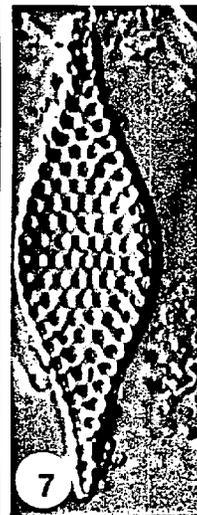
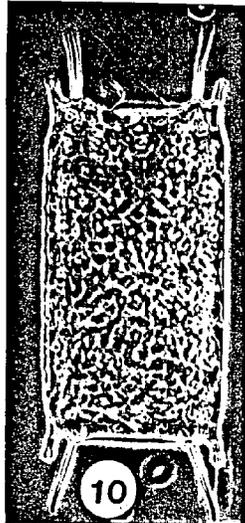
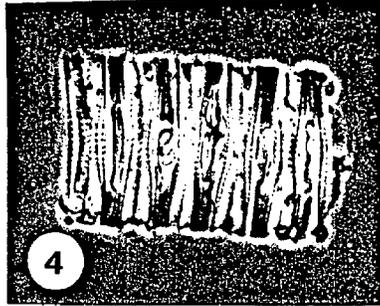
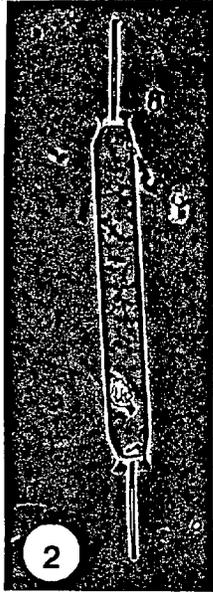
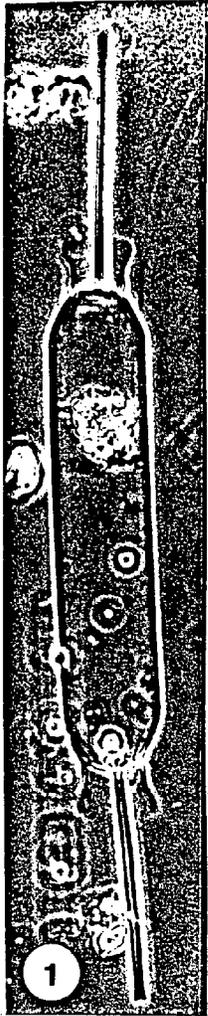


Plate 27

- Fig. 1: *Lithodesmium undulatum* Ehr.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Figs. 2,3: *Biddulphia regia* (Schulze) Ostenfeld
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 4: *Eupodiscus radiatus* Bailey.
FSU Marine Lab., north Florida, Station 00.
04/85. x500.
- Fig. 5: *Aulacodiscus argus* (Ehr.) A. S.
FSU Marine Lab., north Florida, Station 00
01/86. x500.
- Fig. 6: *Cymatosira lorenzianus* Grunow.
FSU Marine lab., north Florida, Station 00.
01/86. x1200.
- Fig. 7: *Pseudauliscus radiatus* (Aul.) Bailey.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 8: *Odontella rhombosa* Ehr.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 9: *Trigonium alternans* (Bailey) Mann.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 10: *Eunatogramma laeve* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 11: *Chaetoceros peruvianus* Brightwell.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.

PLATE 27

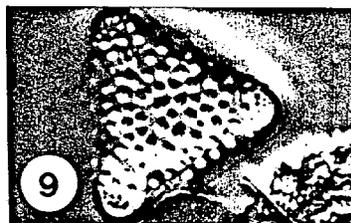
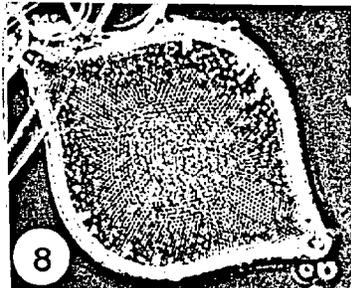
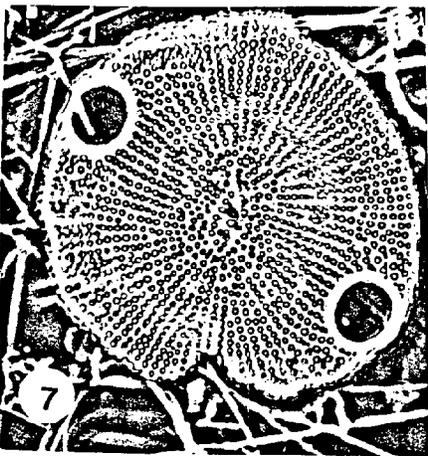
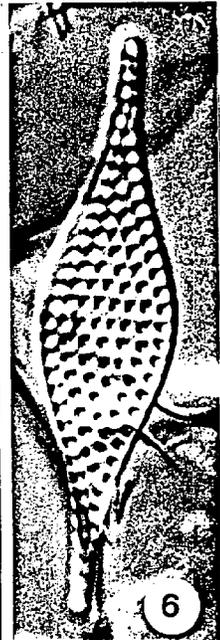
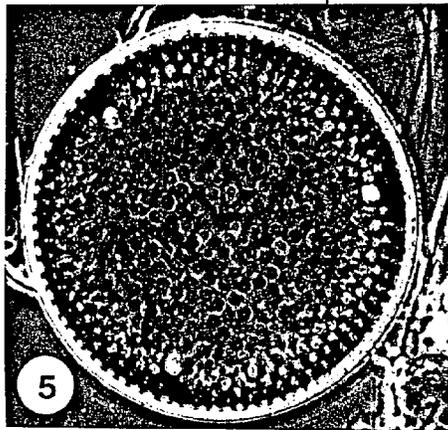
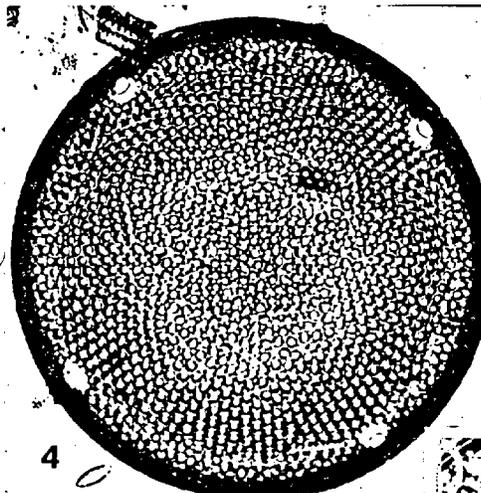
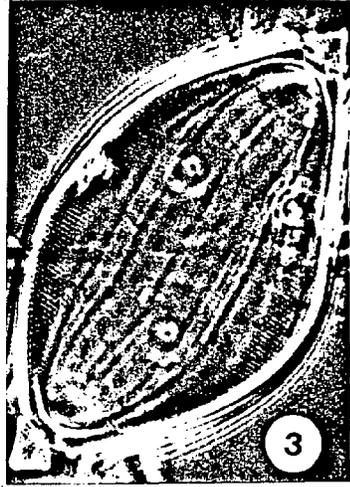
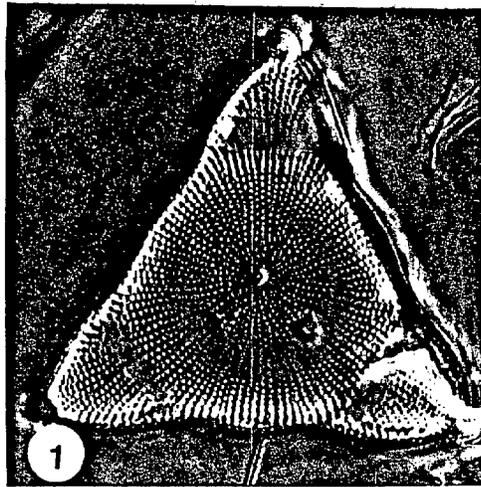


Plate 28

- Fig. 1: *Eucampia zoodiacus* Ehr.
Apalachee Bay, north Florida, Station A-03.
10/87. x250.
- Figs. 2,3: *Eucampia cornuta* (Cleve) Grunow.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Figs. 4,5: *Streptotheca themensis* Shrubbs.
Apalachee Bay, north Florida, Station A-01.
10/87. x250.
- Fig. 6: *Eucampia zoodiacus* Ehr.
FSU Marine Lab., north Florida, Station 00.
01/86. x500.
- Fig. 7: *Eucampia cornuta* (Cl.) Grunow.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 8: *Eucampia zoodiacus* Ehr.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 9: *Eucampia zoodiacus* Ehr.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x500.
- Fig. 10: *Eucampia cornuta* (Cl.) Grunow.
Choctawhatchee bay, north Florida, Station 35.
10/86. x250.
- Fig. 11 *Hemiaulus membranaceus* Cleve.
Choctawhatchee Bay, north Florida, Station C-03.
10/87. x500.
- Figs. 12,13: *Eucampia zoodiacus* Ehr.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.

PLATE 28

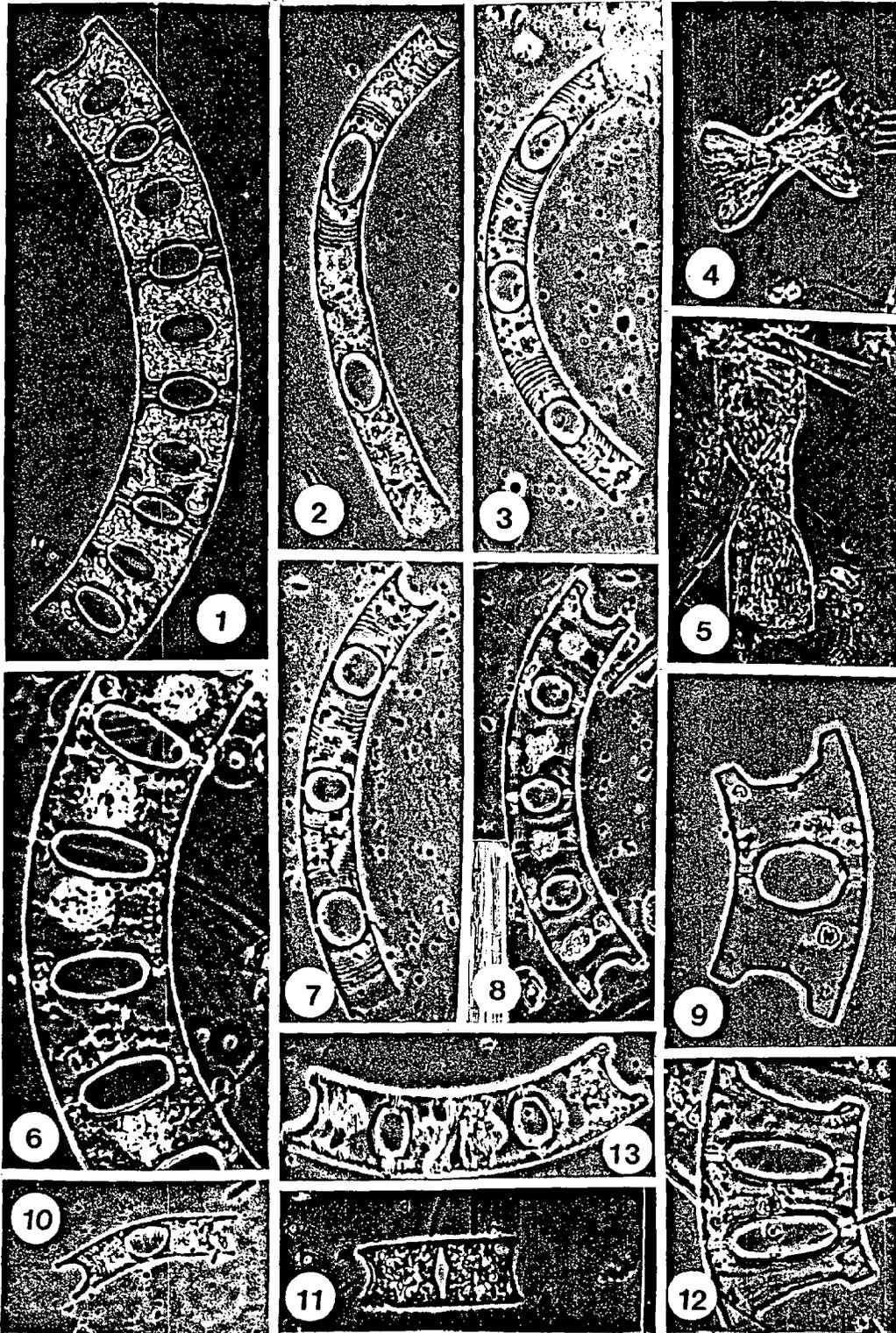


Plate 29

- Figs 1,2: *Grammatophora oceanica* (Ehr.) Grunow.
Choctawhatchee Bay, north Florida, Station 04.
09/85. Fig. 1: x500. Fig. 2: x250.
- Fig.3: *Rhabdonema adriaticum* Kuetz.
Choctawhatchee Bay, north Florida, Station 04.
09/85. x250.
- Fig. 4: *Grammatophora oceanica* (Ehr.) Grunow.
Choctawhatchee Bay, north Florida, Station 04.
09/85. x500.
- Figs. 5,6: *Rhabdonema adriaticum* Kuetz.
Choctawhatchee Bay, north Florida, Station 38.
09/86. x250.
- Fig. 7,8,9: *Striatella unipunctata* (Lyngb.) Agardh.
Choctawhatchee Bay, north Florida, Station 38.
12/85. x500.
- Fig. 10: *Opephora martyi* Heribaud.
Choctawhatchee Bay, north Florida, Station 25.
10/85. x1200.
- Fig. 11: *Dephineis surirella* (Ehr.) Andrews.
Choctawhatchee Bay, north Florida, Station 36.
12/85. x1200.
- Fig. 12: *Striatella unipunctata* (Lyngb.) Agardh.
Choctawhatchee Bay, north Florida, Station 31.
12/85. x1200.

PLATE 29

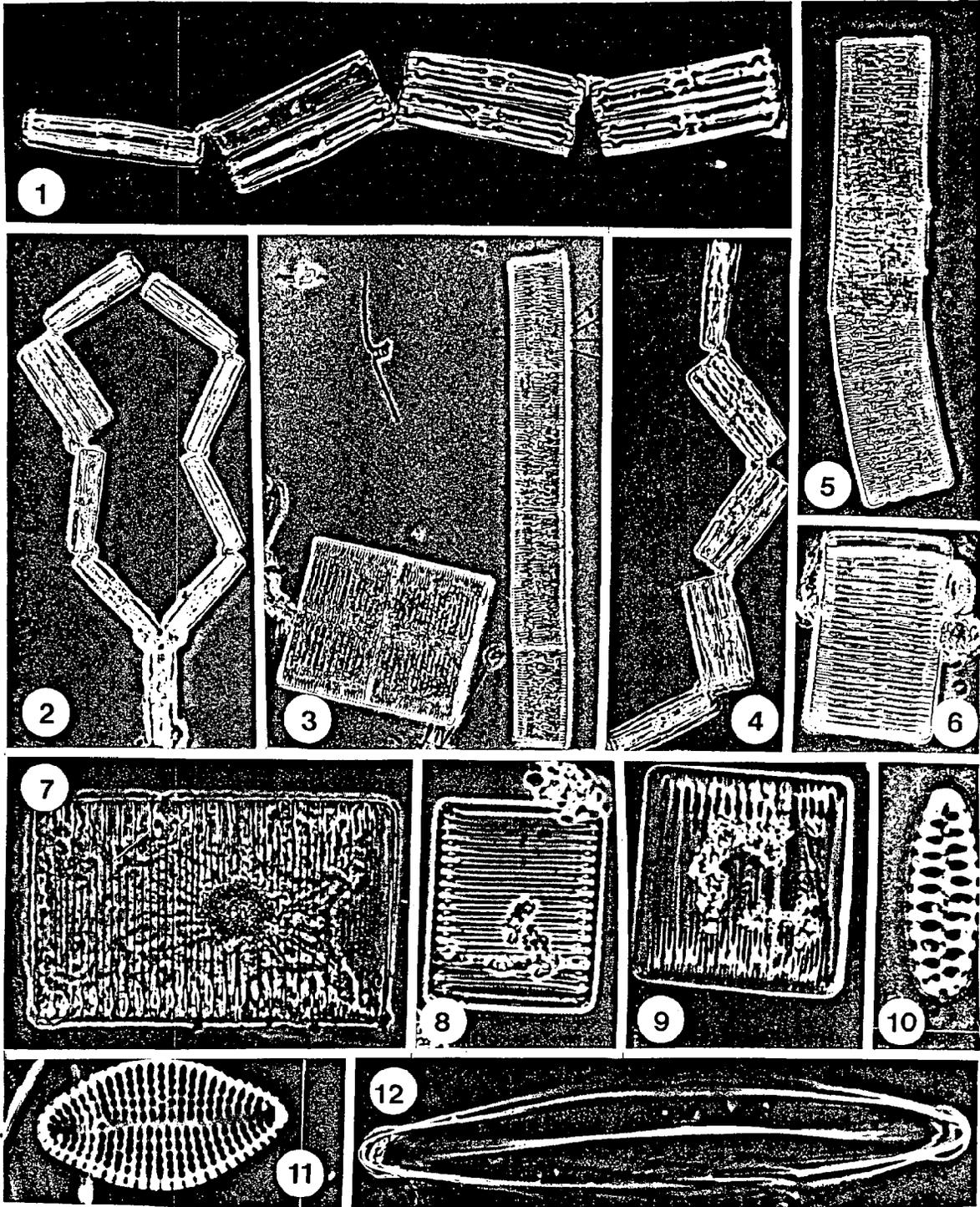


Plate 30

- Figs. 1-5: *Neodelphineis pelagica* Takano.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 6: *Delphineis surirella* (Ehr.) Andrews.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 7: *Rhaphoneis ampiceros* v. *gemmifera* (Ehr.) Hust.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Figs. 8,9: *Opephora pacifica* (Grun.) Petit.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Figs. 10,17: *Delphineis livingstonii* Prasad.
Apalachicola Bay, north Florida, Station E-01.
02/87. x1200.
- Fig. 11: *Perissonoe crucifera* (Kitton) Desik. et al.,
Apalachicola Bay, north Florida, Station E-02.
02/87. x1200.
- Fig. 12: *Dimeregramma marinum* (Greg.) Ralfs.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200
- Fig. 13: *Dimeregramma furcigerum* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 14: *Dimeregramma fulvum* (Greg.) Ralfs.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 15: *Dimeregramma minor* (Greg.) Ralfs.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 16: *Thalassionema nitzschioides* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 18: *Plagigramma staurophorum* (Greg.) Heiberg.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.

PLATE 30

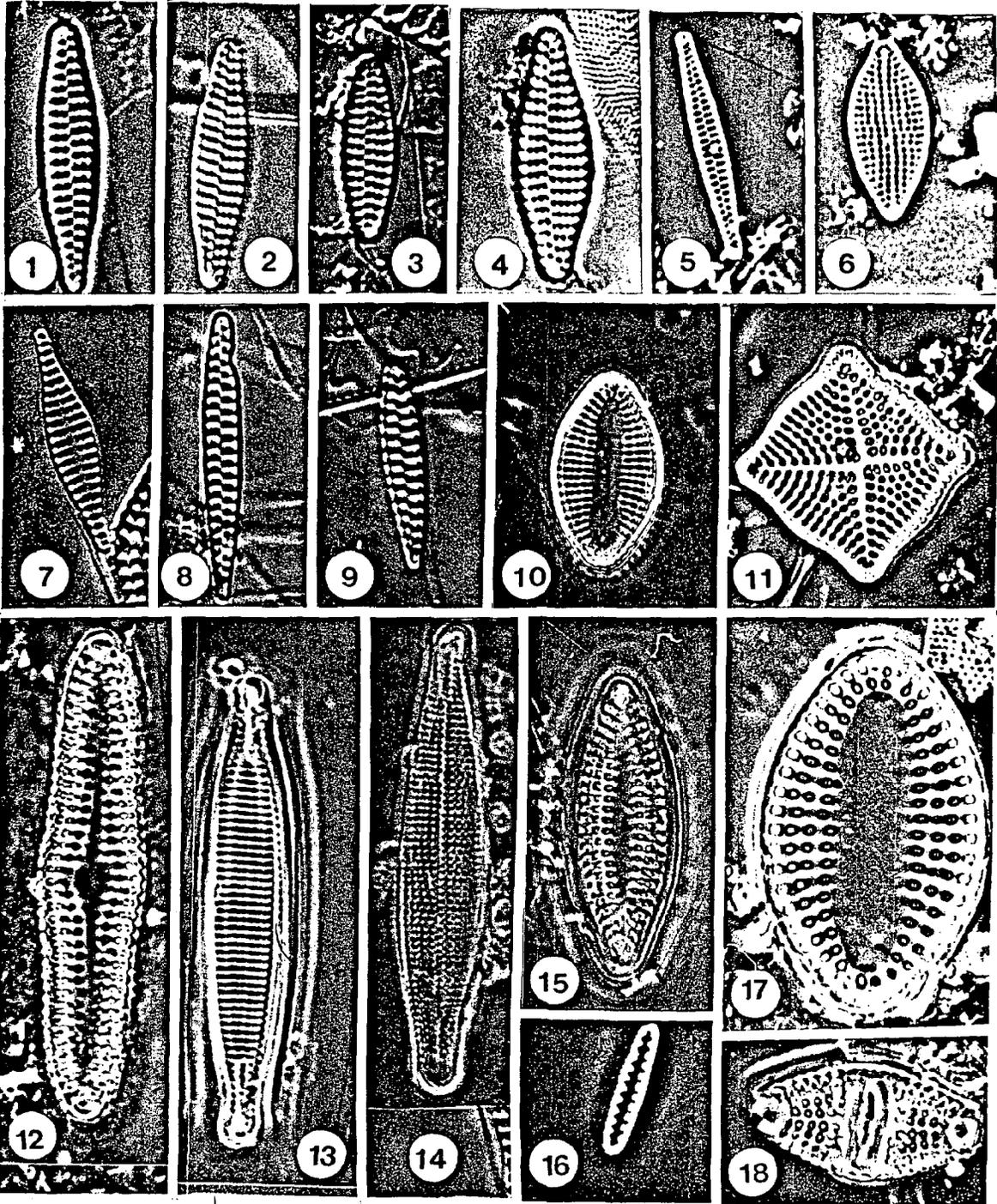


Plate 31

- Fig. 1: *Synedra pulchella* (Ralfs) Kuetz.
FSU Marine Lab., north Florida, Station 00.
01/86. x500.
- Fig. 2: *Thalassiothrix longissima* Cleve & Grunow.
Choctawhatchee Bay, north Florida, Station 15.
03/86. x200.
- Figs. 3,8: *Thalassiothrix mediterranea* v. *pacifica* Cupp.
Choctawhatchee Bay, north Florida, Station 19.
02/86. x1000.
- Fig. 4: *Synedra tabulata* (Ag.) Kuetz.
Choctawhatchee Bay, north Florida, Station 22.
01/85. x1000.
- Fig. 5: *Licmophora abbreviata* Agardh.
Apalachee Bay, north Florida, Station A-03.
10/97. x1000.
- Fig. 6: *Navicula forcipata* Greville.
Choctawhatchee Bay, north Florida, Station 22.
01/85. x1000.
- Fig. 7: *Fragilaria leptopstuaron* v. *rhomboides* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x1000.
- Fig. 9: *Asterionella japonica* Cleve.
FSU Marine Lab., north Florida, Station M-03.
10/87. x1000.
- Fig. 10: *Striatella unipunctata* (Lyngb.) Agardh.
Choctawhatchee Bay, north Florida, Station 34.
01/86. x1200.

PLATE 31

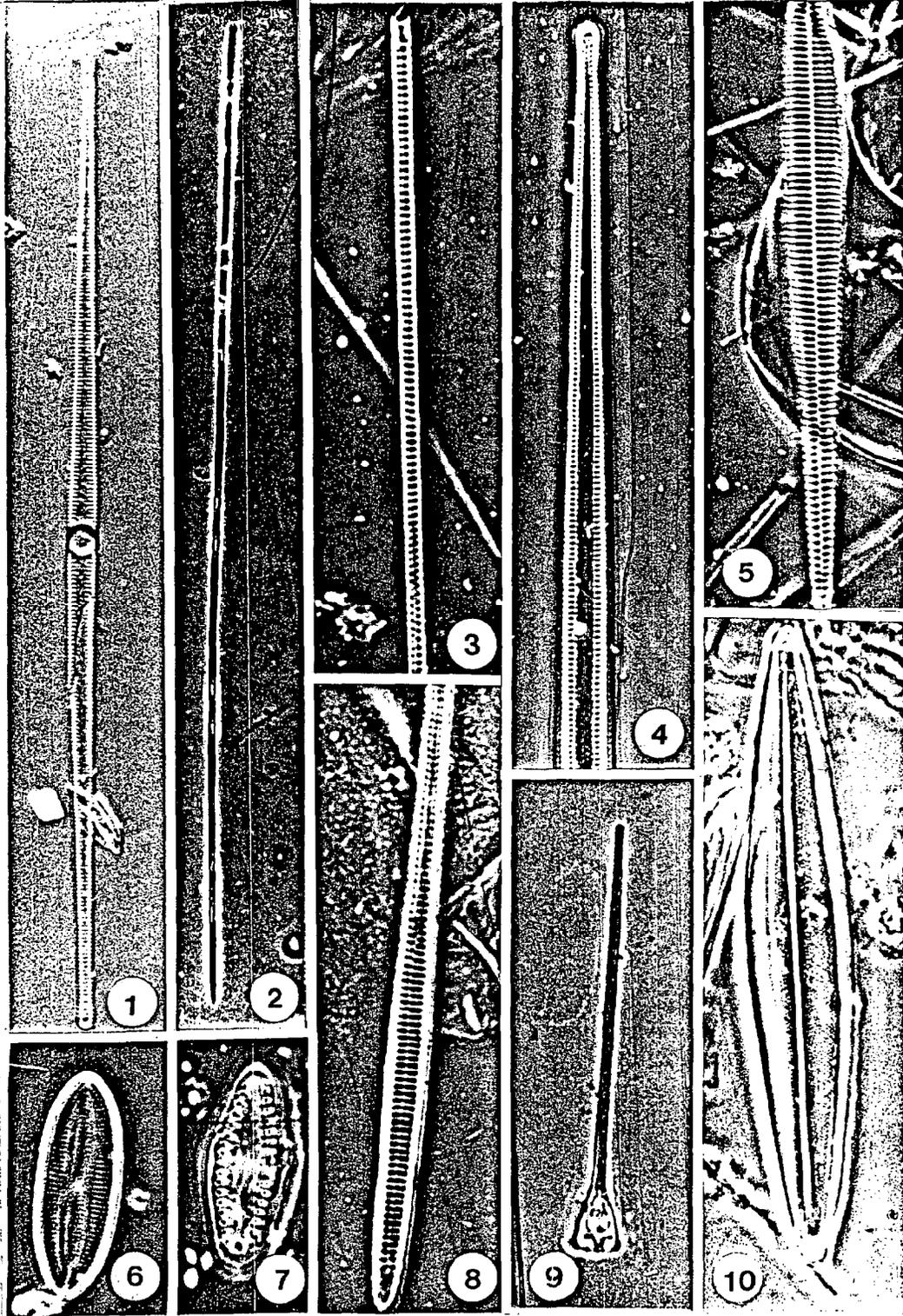


Plate 32

- Fig. 1: *Thalassiothrix longissima* Cleve & Grunow.
Choctawhatchee Bay, north Florida, Station 22.
03/86. x500.
- Fig. 2: *Synedra pulchella* (Ralfs) KUetz.
Apalachee Bay, north Florida, Station A-02.
02/87. x1000.
- Figs. 3,4,5: *Thalassiothrix frauenfeldii* Grunow.
Apalachee Bay, north Florida, Station A-01.
02/88. x500.
- Fig. 6: *Thalassiothrix longissima* Cleve & Grunow.
Choctawhatchee Bay, north Florida, Station 22.
03/86. x500.
- Fig. 7: *Bacillaria paxillifer* (Mueller) Hendey.
Choctawhatchee Bay, north Florida, Station 03.
06/87. x500.
- Figs. 8,9: *Thalassionema nitzschioides* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x500.
- Fig. 10: *Thalassiothrix frauenfeldii* Grunow.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x500.
- Fig. 11: *Asterionella japonica* Cleve.
Apalachee Bay, north Florida, Station A-02.
02/88. x500.

PLATE 32

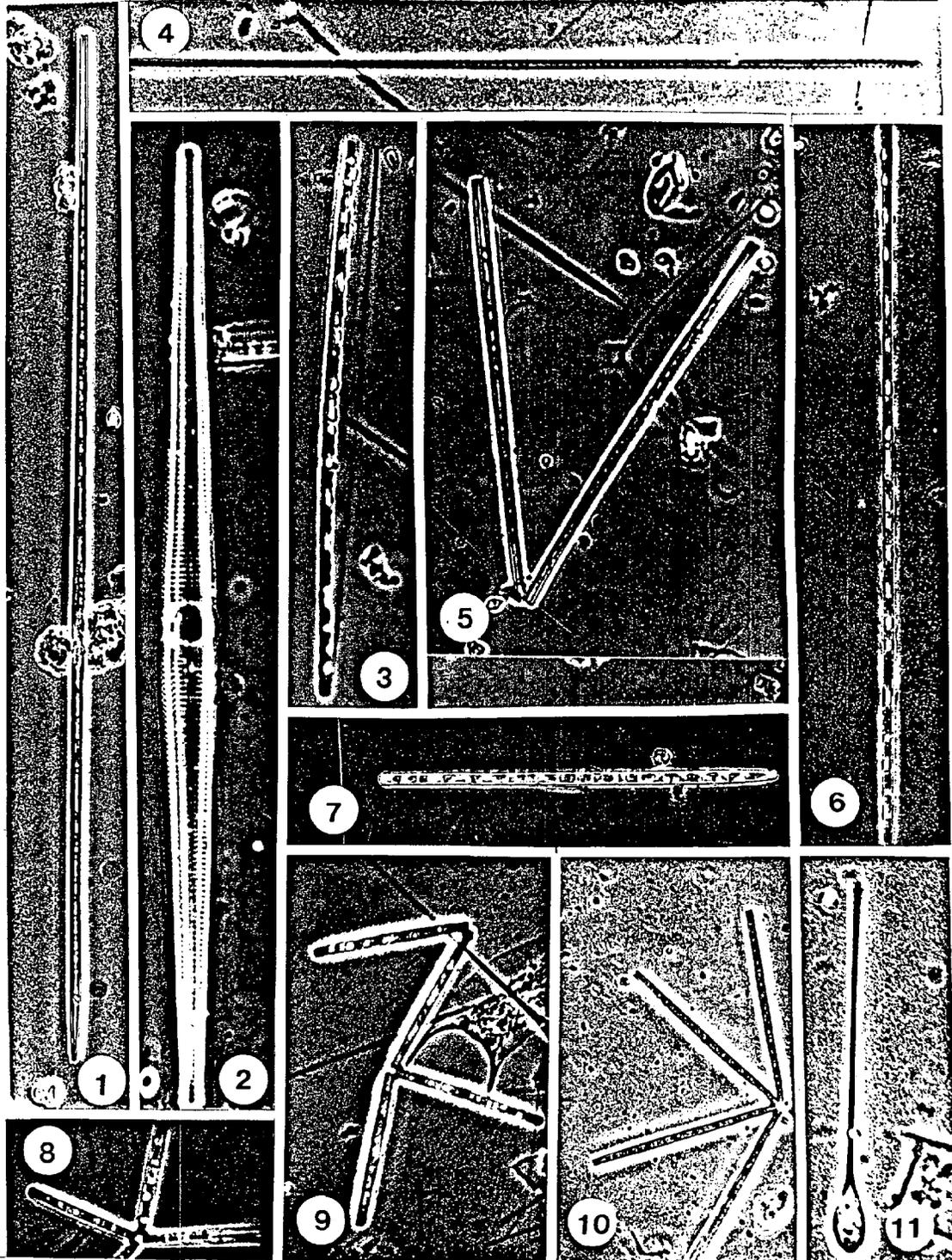


Plate 33

- Figs. 1-4: *Achnanthes manifera* Brun.
Choctawhatchee Bay, north Florida, Station 38.
08/86. x1200.
- Fig. 5: *Diploneis parma* Cleve.
Choctawhatchee Bay, north Florida, Station 03.
10/85. x1200.
- Fig. 6: *Navicula finmarchica* Cleve.
Choctawhatchee Bay, north Florida, Station 19.
10/85. x1200.
- Fig. 7: *Caloneis oregonica* (Ehr.) Patr.
Choctawhatchee Bay, north Florida, Station 03.
10/85. x1200.
- Fig. 8: *Amphora cf. exornata* Grunow.
Choctawhatchee Bay, north Florida, Station 36.
10/85. x1200.
- Fig. 9: *Amphora cf. clara* A. S.
Choctawhatchee Bay, north Florida, Station 38.
08/86. x1200.
- Fig. 10: *Navicula forcipata* Greville.
Choctawhatchee bay, north Florida, Station 34.
10/85. x1200.
- Fig. 11: *Amphora clevei* Grunow.
Choctawhatchee Bay, north Florida, Station 25.
110/85. x1200.
- Fig. 12: *Amphora laevis* Gregory.
Choctawhatchee Bay, north Florida, Station 38.
03/86. x1200.
- Fig. 13: *Amphora proteus* Gregory.
Choctawhatchee Bay, north Florida, Station 34.
03/86. x1200.
- Fig. 14: *Amphora ocellata* Donkin.
Apalachee Bay, north Florida, Station A-02.
02/88. x1200.
- Fig. 15: *Campylodiscus limbatus* Breb.
Choctawhatchee Bay, north Florida, Station 25.
04/86. x500.
- Fig. 16: *Amphora coffeaeformis* (Ag.) Kuetz.
Choctawhatchee Bay, north Florida, Station 36.
12/85. x1200.

PLATE 33

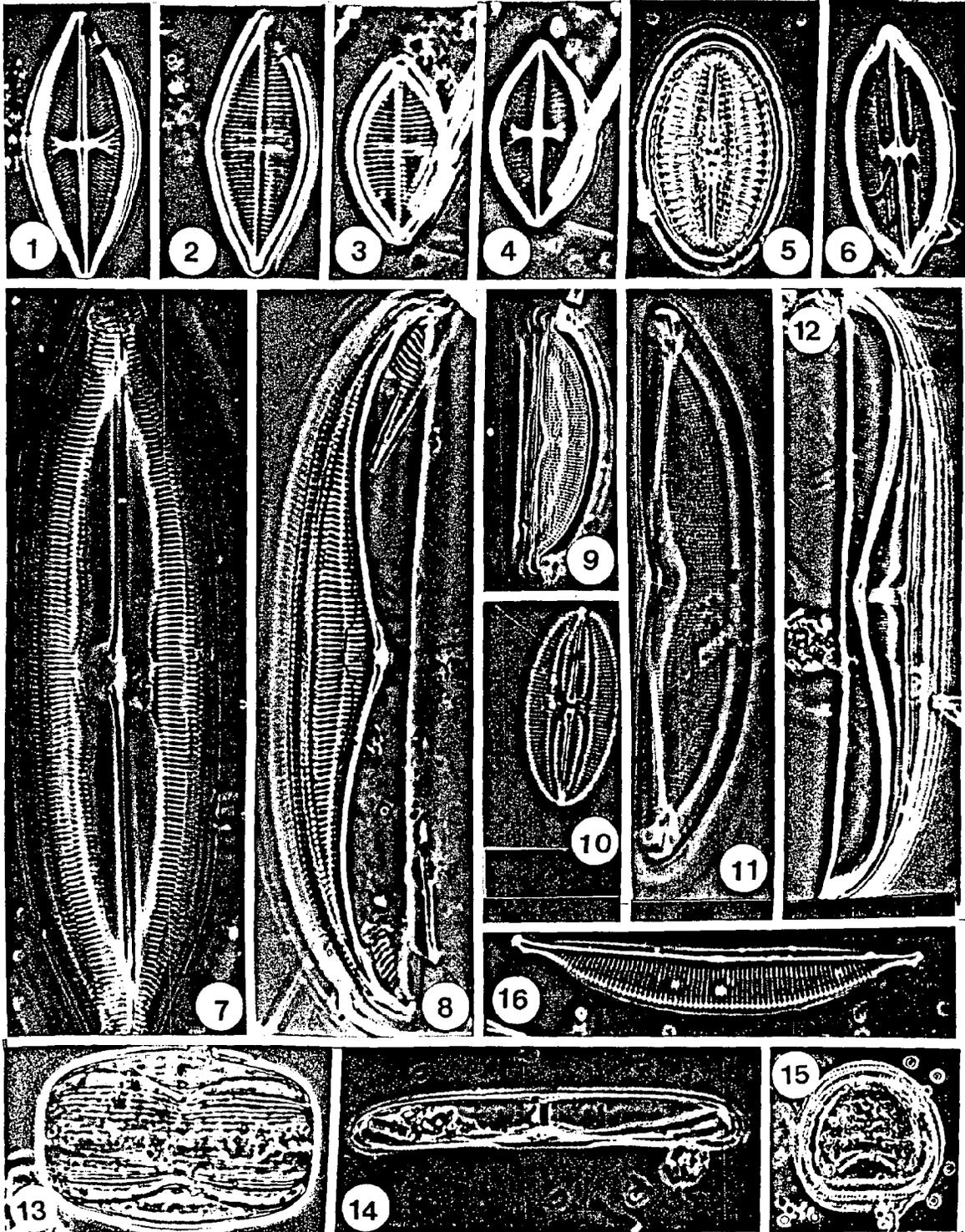


Plate 34

- Figs. 1,2: *Achnanthes citronella* (Mann) Hustedt.
Choctawhatchee Bay, north Florida, Station 22.
04/86. x1200.
- Fig. 3: *Cocconeis decipiens* Cleve.
Choctawhatchee Bay, north Florida, Station 03.
02/86. x1200.
- Fig. 4: *Campyloneis grevillei* (W. Sm.) Grun.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Figs. 5,6: *Mastogloia cf. elliptica* (Ag.) Cleve.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Figs. 7,8: *Mastogloia cf. subacuta* Hustedt.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Figs. 9,14: *Mastogloia ignorata* Hustedt.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Figs.10,11: *Dictyoneis marginata* (Lewis) Cleve.
Apalachicola Bay, north Florida, Station E-01.
02/87. x1200.
- Figs.12,13: *Mastogloia cf. baldjikiana* Grunow.
Choctawhatchee Bay, north Florida, Station 35.
05/87. x1200.

PLATE 34

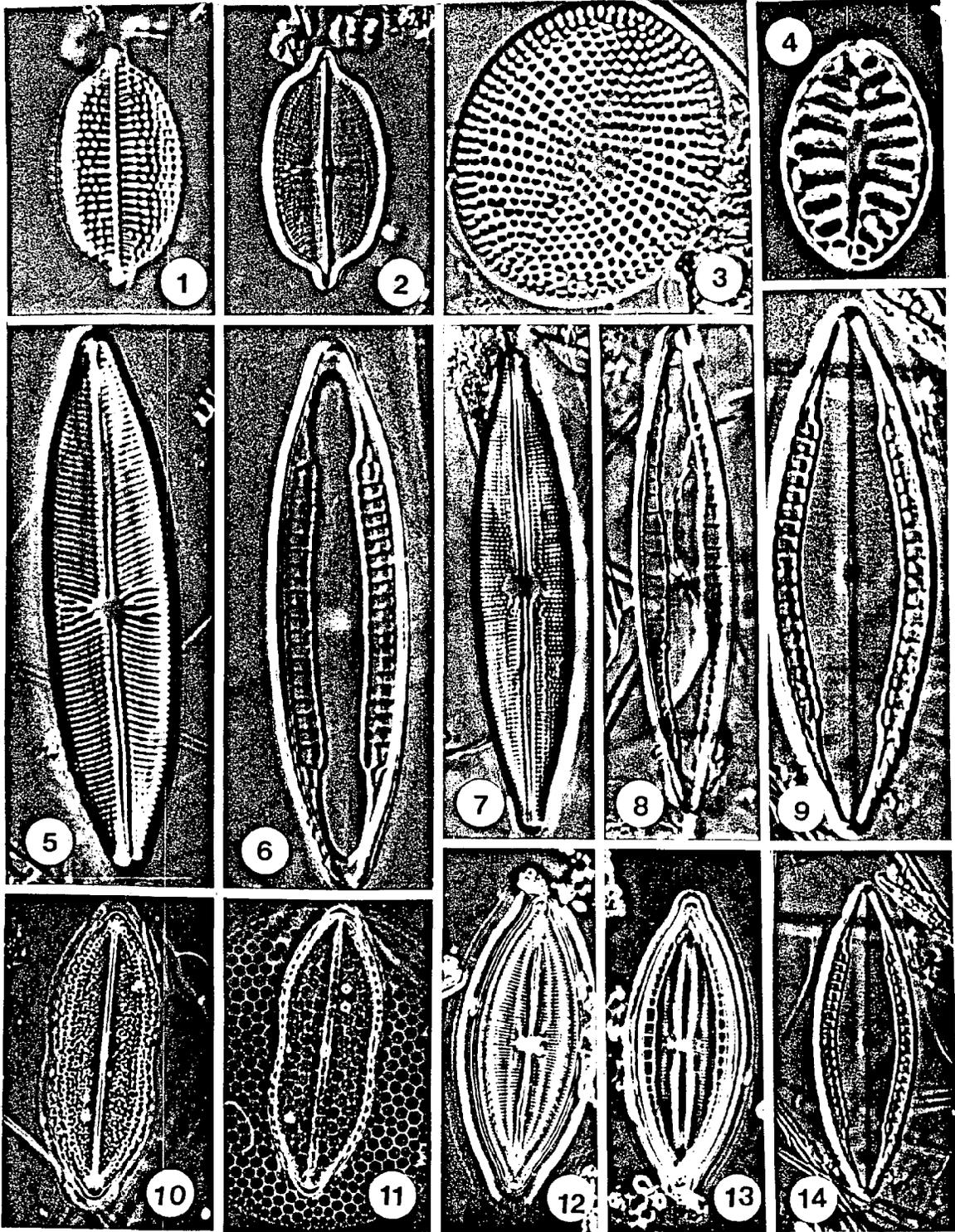


Plate 35

- Figs. 1,2: *Mastogloia cf. elliptica* (Ag.) Cleve.
Choctawhatchee Bay, north Florida, Station 03.
05/87. x1200.
- Figs. 3,4: *Mastogloia braunii* Grunow.
Choctawhatchee Bay, north Florida, Station 31.
08/86. x1200.
- Figs. 5,10: *Mastogloia erythraea* Grunow.
Choctawhatchee Bay, north Florida, Station 38.
07/86. x1200.
- Figs. 6,7: *Mastogloia cf. elegans* Lewis.
Choctawhatchee Bay, north Florida, Station 38.
08/86. x1200.
- Figs. 8,9: *Mastogloia baldjikiana* Grunow.
Choctawhatchee Bay, north Florida, Station 38.
07/86. x1200.
- Fig. 11: *Navicula lyra v. insignis* A. S.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 12: *Navicula diffluens* A. S.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 13: *Navicula menaiana* Hendey.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 14: *Navicula abunda* Hustedt.
FSU Marine Lab., north Florida, Station 00
01/86. x1200.

PLATE 35

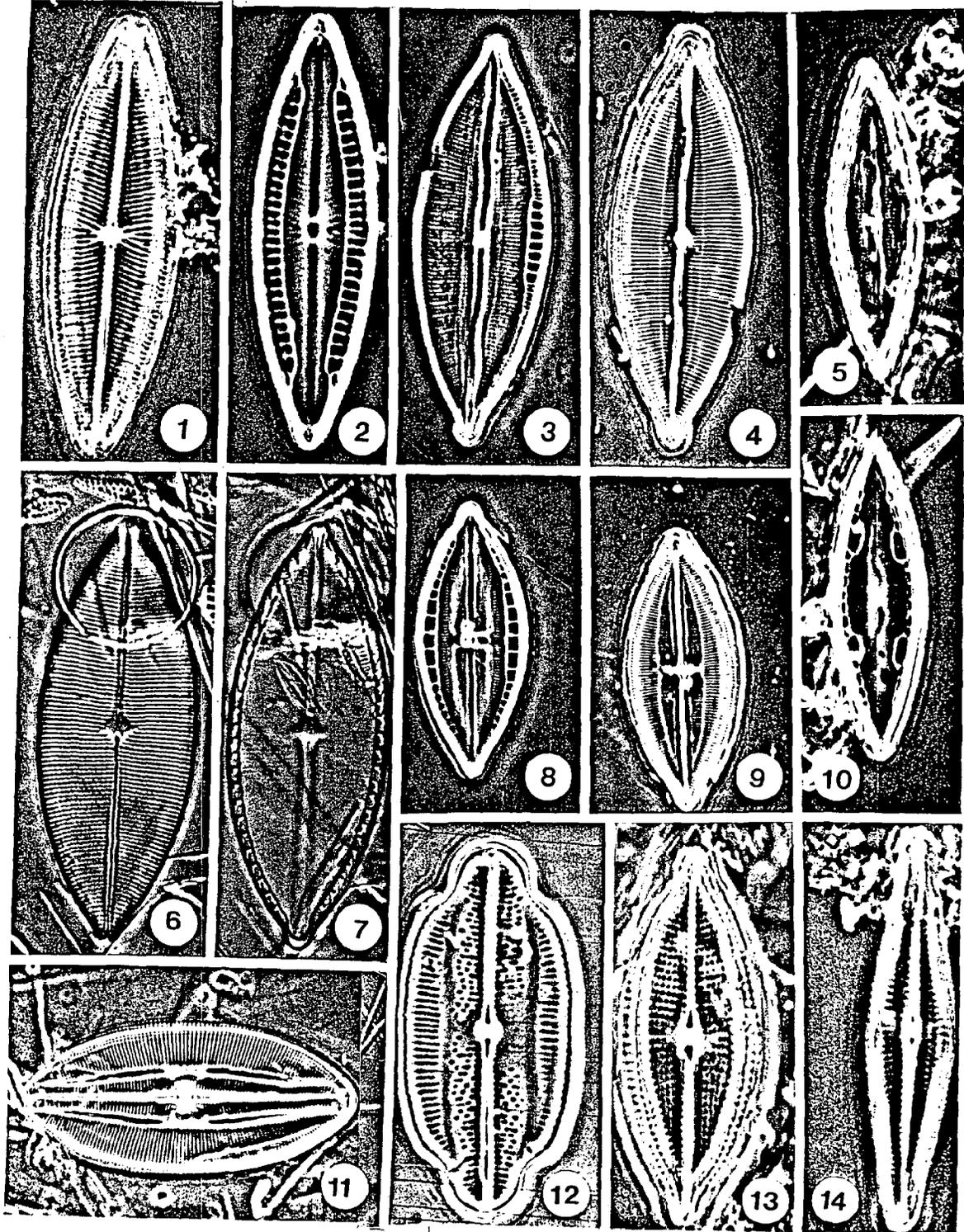


Plate 36

- Figs. 1,2: *Mastogloia smithii* Thwaites.
Choctawhatchee Bay, north Florida, Station 22.
10/85. x1200.
- Fig. 3: *Mastoneis biformis* (Grun.) Cleve.
Choctawhatchee Bay, north Florida, Station 36.
02/86. x1200.
- Fig. 4: *Mastogloia angulata* Lewis.
Choctawhatchee Bay, north Florida, Station 19.
11/85. x1200.
- Fig. 5: *Mastogloia* sp.
Choctawhatchee Bay, north Florida, Station 38.
10/85. x1200.
- Fig. 6: *Navicula cf. granulata* Breb.
Choctawhatchee Bay, north Florida, Station 03.
08/87. x1200.
- Fig. 7: *Navicula marina* Ralfs.
Choctawhatchee Bay, north Florida, Station 15.
08/87. x1200.
- Figs. 8,9: *Mastogloia portierana* A. S.
Choctawhatchee Bay, north Florida, Station 03.
03/86. x1200.
- Figs. 10,11: *Mastogloia angulata* Lewis.
Choctawhatchee Bay, north Florida, Station 38.
08/86. x1200.
- Fig. 12: *Navicula cf. pseudoscutiformis* Hustedt.
Choctawhatchee Bay, north Florida, Station 03.
10/86. x1200.
- Fig. 13: *Navicula lyra* Ehr.
Choctawhatchee Bay, north Florida, Station 03.
11/87. x1200.
- Fig. 14: *Navicula cf. pygmaea* Kuetz.
Choctawhatchee Bay, north Florida, Station 03.
11/87. x1200.

PLATE 36

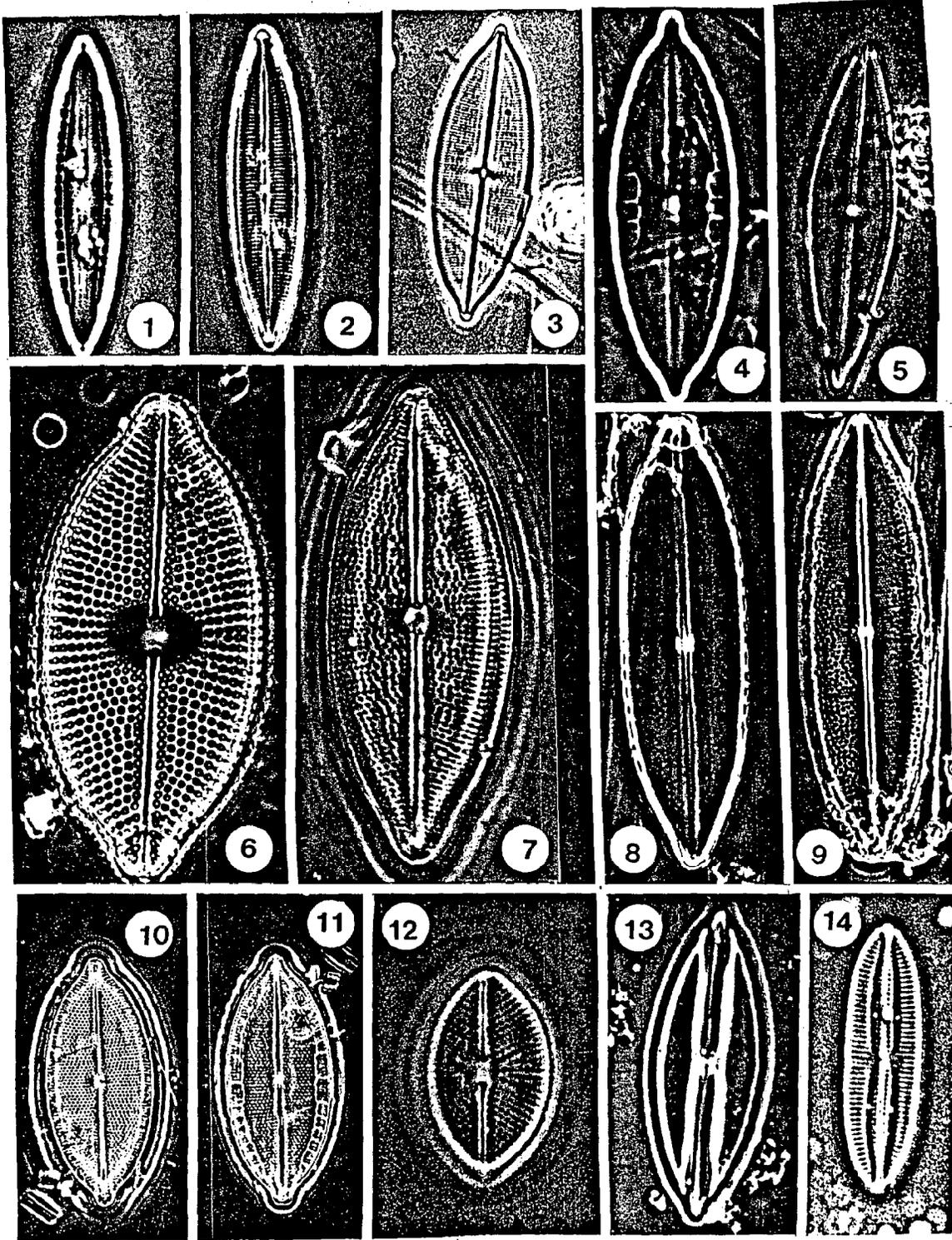


Plate 37

- Fig. 1: *Mastogloia foliolum* Brun in A.S.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x500.
- Fig. 2: *Navicula lyra* Ehr.
Choctawhatchee Bay, north Florida, Station 15.
08/787. x500.
- Fig. 3: *Navicula pennata* Schmidt.
Choctawhatchee Bay, north Florida, Station 03.
04/87. x1200.
- Figs. 4,6: *Gyrosigma fasciola* (Ehr.) Griffith & Henfrey.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x1200.
- Fig. 5,14: *Haslea* sp.
Choctawhatchee Bay, north Florida, Station 35.
10/86. Fig. 5: x250. Fig. 14: x500.
- Fig. 7: *Navicula normalis* A. S.
Choctawhatchee Bay, north Florida, Station 34.
10/85. x1200.
- Figs. 8,9: *Pleurosigma strigosum* Wm. Smith.
Choctawhatchee Bay, north Florida, Station 38.
08/86. Fig. 8: x500. Fig. 9: x1200.
- Fig. 10: *Navicula* sp.
Choctawhatchee Bay, north Florida, Station 03.
11/87. x 1200.
- Fig. 11: *Navicula* sp.
Choctawhatchee Bay, north Florida, Station 38.
08/86. x1200.
- Fig. 12: *Navicula abunda* Hustedt.
Choctawhatchee Bay, north Florida, Station 15.
08/86. x1200.
- Fig. 13: *Diploneis crabro* Ehr.
Choctawhatchee Bay, north Florida, Station 19.
11/85. x1200.

PLATE 37

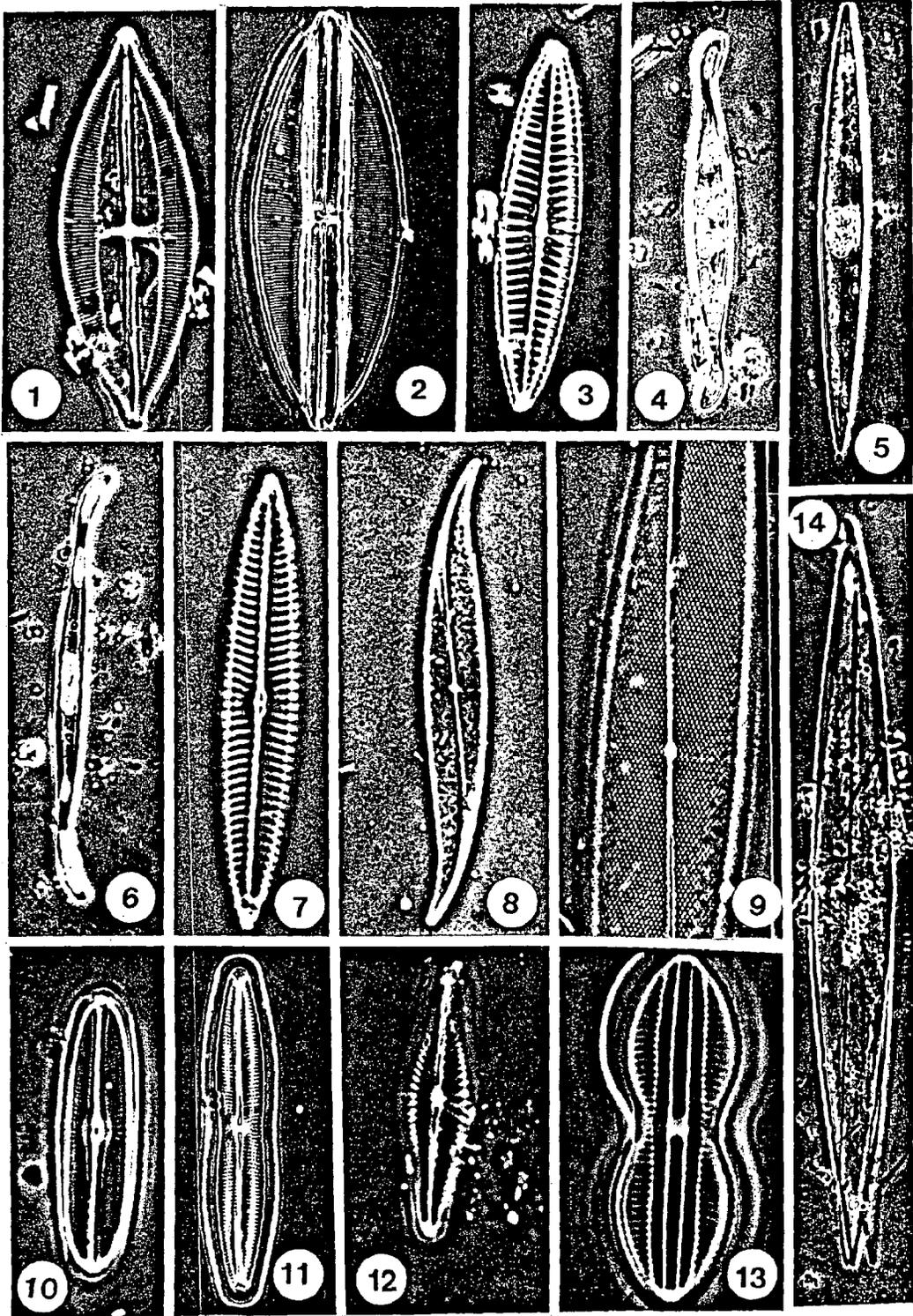


Plate 38

- Fig. 1: *Amphiprora alata* (Ehr.) Kuetz.
Choctawhatchee Bay, north Florida, Station 03.
01/86. x500.
- Fig. 2: *Diploneis cf. reichardtii* Heiden.
Choctawhatchee Bay, north Florida, Station 03.
01/86. x1200.
- Fig. 3: *Navicula spectabilis* Greg.
Choctawhatchee Bay, north Florida, Station 07.
01/86. x1200.
- Fig. 4: *Navicula cruciculoides* Brockmann.
Choctawhatchee Bay, north Florida, Station 15.
02/86. x1200.
- Fig. 5: *Navicula clavata* Gregory.
Choctawhatchee Bay, north Florida, Station 22.
03/86. x1200.
- Fig. 6: *Mastogloia foliolum* Brrun in Schmidt.
Choctawhatchee Bay, north Florida, Station 38.
09/85. x1200.
- Fig. 7: *Navicula forcipata* Greville.
Choctawhatchee Bay, north Florida, Station 38.
03/86. x1200.
- Figs. 8,9: *Gyrosigma fasciola v. arcuata* (Donkin) Cl.
Choctawhatchee Bay, north Florida, Station 22.
04/86. x1200.
- Fig. 10. *Mastogloia baldjikiana* Grunow.
Choctawhatchee Bay, north Florida, Station 38.
07/86. x1200.
- Fig. 11: *Diploneis cf. weissflogii* (A. S.) Cleve.
FSU Marine Lab., north Florida, Station 00.
04/85. x1200.
- Fig. 12: *Navicula cf. maculosa* Donk.
FSU Marine Lab., north Florida, Station 00.
04/85. x1200.

PLATE 38

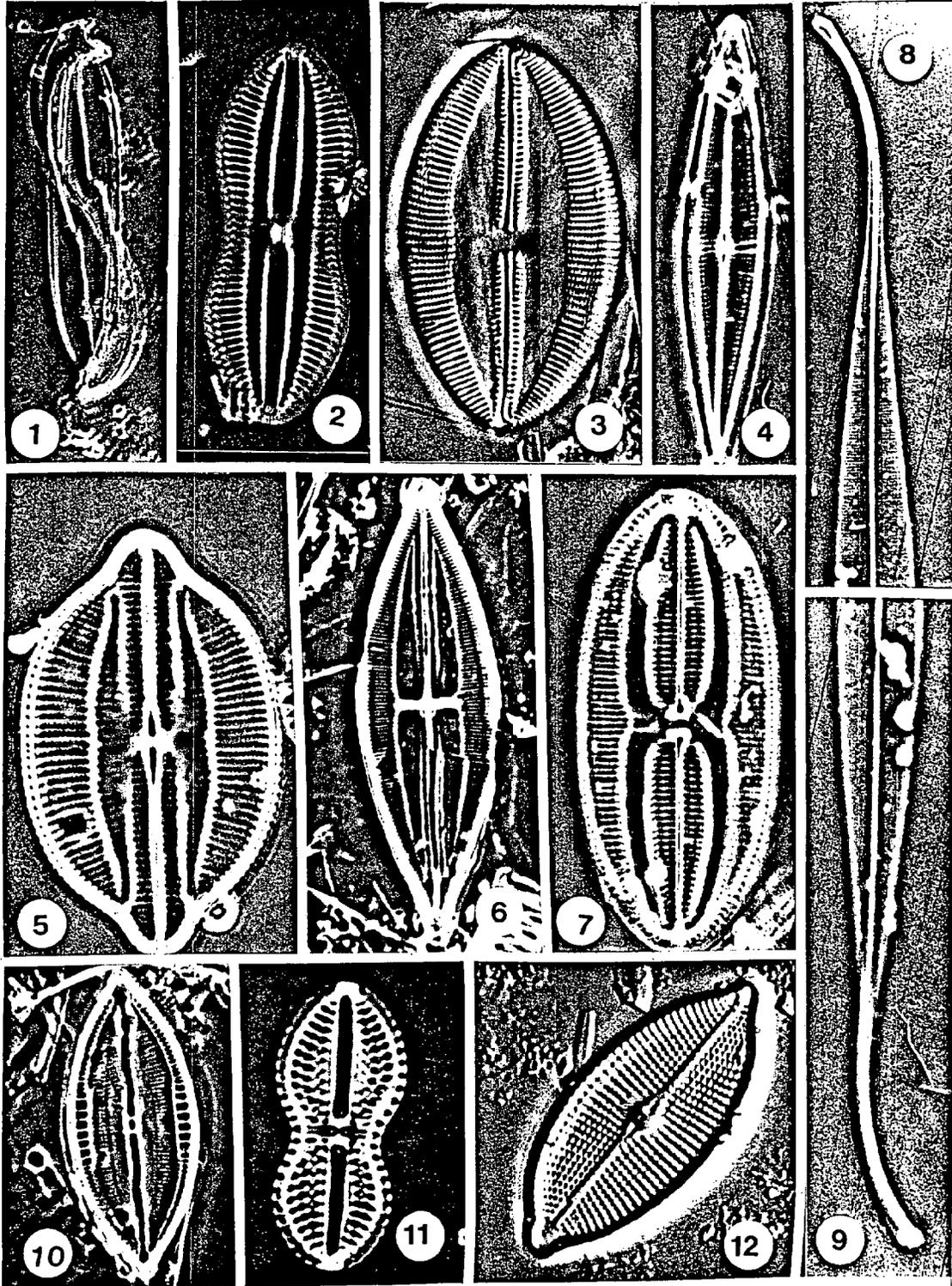


Plate 39

- Fig. 1: *Cymbella* sp.
Choctawhatchee Bay, north Florida, Station 07.
11/87. x1200.
- Figs. 2,3: *Mastogloia euxina* Cleve.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 4: *Amphora* sp.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 5: *Navicula peregrina* (Ehr.) Kuetz.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 6: *Navicula* sp.
Choctawhatchee Bay, north Florida, Station 38.
12/87. x1200.
- Fig. 7: *Amphora hosatica* Hustedt.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 8: *Amphora binoides* var. *interrupta* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 9: *Navicula* cf. *nummularia* Greville.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 10: *Navicula directa* v. *remota* Cleve.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 11: *Navicula clamans* Cl.
FSU Marine Lab., north Florida Station 00.
01/86. x1200.

PLATE 39

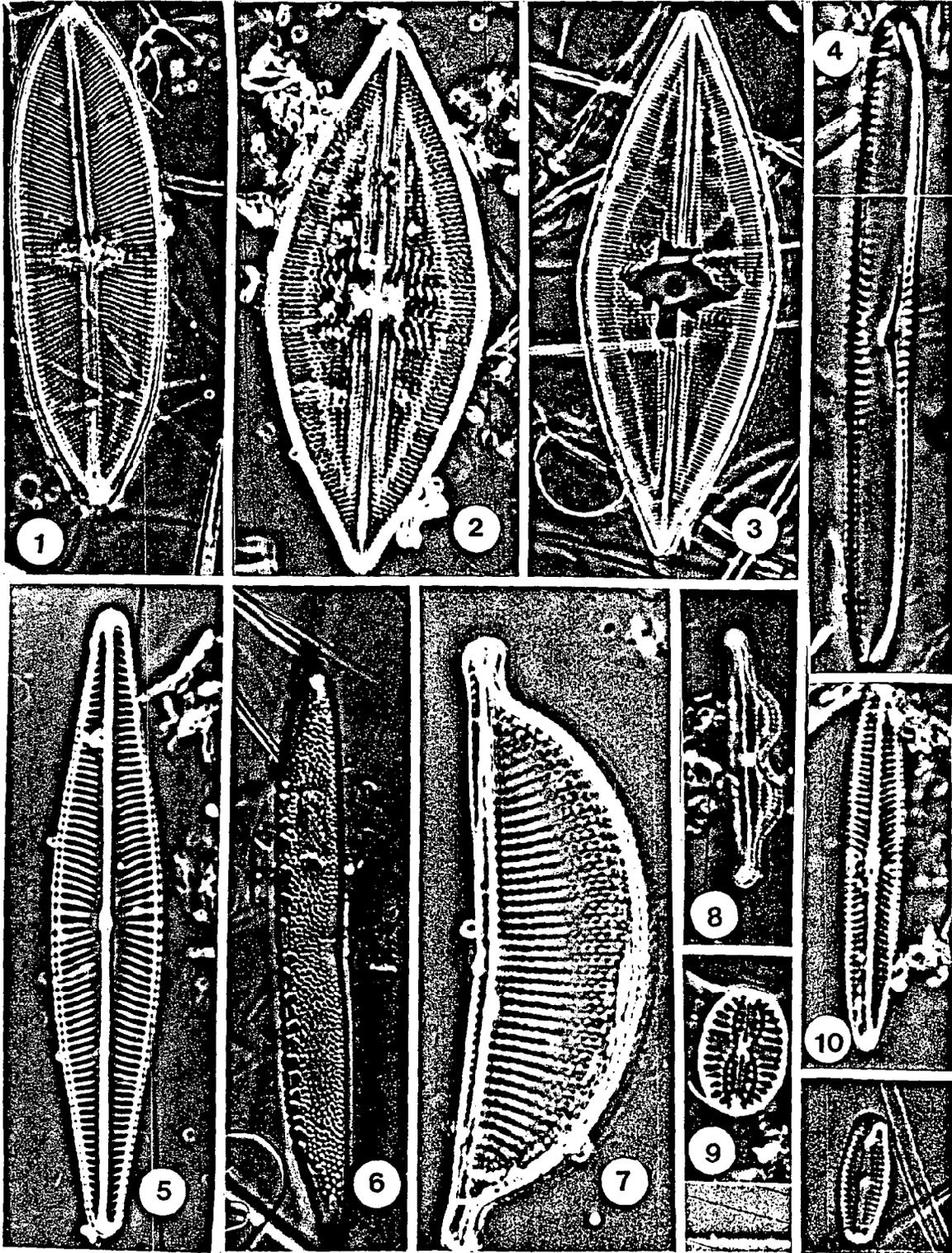


Plate 40

- Fig. 1: *Navicula* sp. " " " " " "
Choctawhatchee Bay, north Florida, Station 03.
05/87. x1200.
- Fig. 2: *Navicula yarrensensis* v. *americana* Cl.
Choctawhatchee Bay, north Florida, Station 03.
05/87. x1200.
- Fig. 3: *Stephanopyxis turris* (Grev. & Arn.) Ralfs.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x1200.
- Fig. 4: *Navicula* cf. *algida* Grunow.
Choctawhatchee Bay, north Florida, Station 03.
05/87. x1200.
- Fig. 5: *Navicula* cf. *tuscula* Grunow.
Choctawhatchee Bay, north Florida, Station 03.
05/87. x1200.
- Fig. 6: *Trigonium arcticum* (Brightwell) Cleve.
Choctawhatchee Bay, north Florida, Station 03.
06/87. x1200.
- Fig. 7: *Navicula granulata* Bailey.
Choctawhatchee Bay, north Florida, Station 03.
06/87. x1200.
- Fig. 8: *Navicula abunda* Hustedt.
Choctawhatchee Bay, north Florida, Station 15.
06/87. x500.
- Fig. 9: *Dinophysis caudata* v. *acutiformis*.
Choctawhatchee Bay, north Florida, Station 36E
05/87. x500.
- Fig. 10: *Exuviella baltica* Lohmann.
Choctawhatchee Bay, north Florida, Station 36E
05/87. x500.

PLATE 40

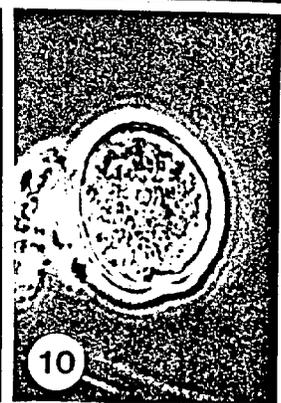
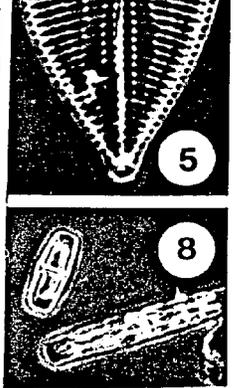
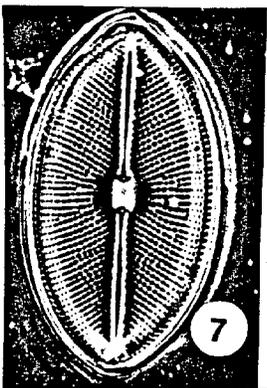
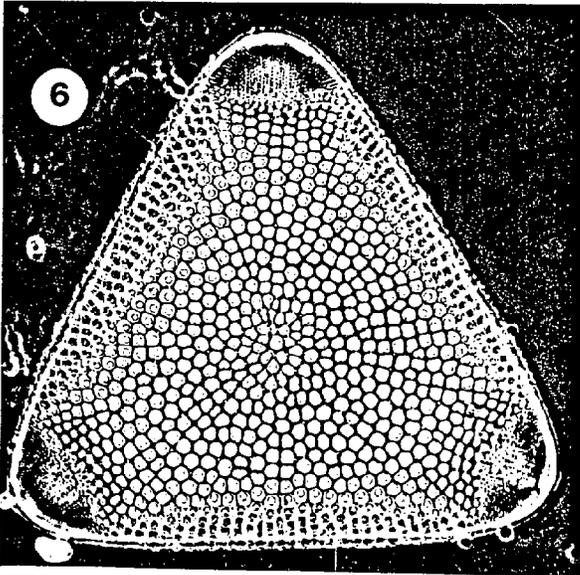
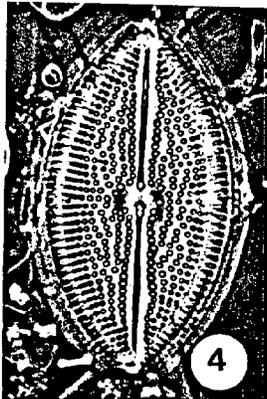
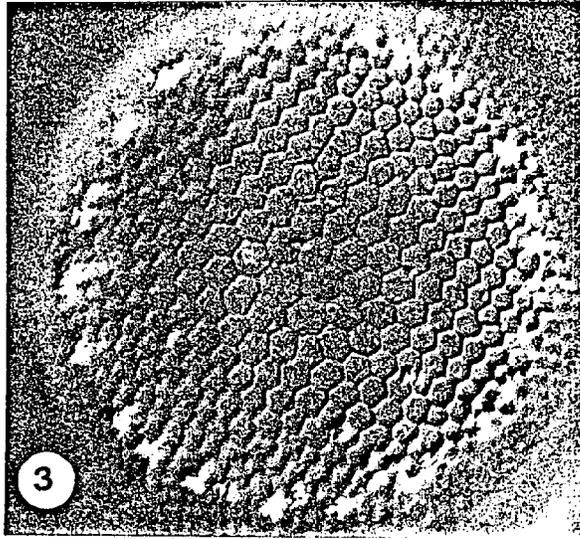
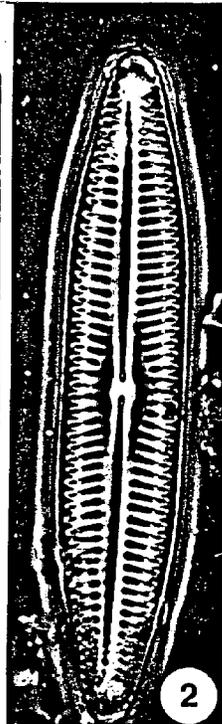
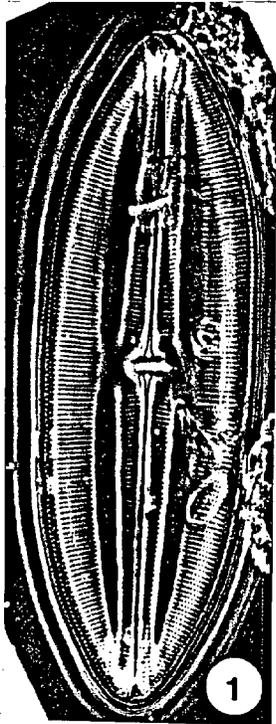


Plate 41

- Fig. 1: *Amphora decussata* Grunow.
Choctawhatchee Bay, north Florida, Station 22.
07/87. x1200.
- Fig. 2: *Amphora cf. graeffi v. minor* Pereg.
Choctawhatchee Bay, north Florida, Station 22.
08/87. x1200.
- Fig. 3: *Amphora ocellata* Donkin.
Choctawhatchee Bay, north Florida, Station 15.
07/87. x1200.
- Fig. 4: *Amphora coffeaeformis* (Ag.) Kuetz.
Apalachee Bay, north Florida, Station A-03.
02/88. x1200.
- Fig. 5: *Nitzschia circumscuta* (Bailey) Grunow.
Choctawhatchee Bay, north Florida, Station 03.
6/87. x500.
- Fig. 6: *Nitzschia hungarica* Grunow.
Choctawhatchee Bay, north Florida, station 03.
06/87. x1000.
- Fig. 7: *Nitzschia longissima* (Breb.) Ralfs.
FSU Marine Lab., north Florida, Station 00.
01/86. x500.
- Fig. 8: *Nitzschia reversa* Wm. Sm.
Choctawhatchee Bay, north Florida, Station 03.
06/87. x500.
- Fig. 9: *Nitzschia closterium* (Ehr.) Wm. Sm.
Choctawhatchee Bay, north Florida, Station 15.
06/87. x500.
- Fig. 10: *Amphora arenaria* Donkin.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 11: *Amphora* sp.
Choctawhatchee Bay, north Florida, Station 15.
05/87. x1200.
- Fig. 12: *Amphora astrearia* Breb.,
Choctawhatchee Bay, north Florida, Station 15.
05/87. x1200.
- Figs. 13, 14: *Navicula abunda* Hustedt.
Choctawhatchee Bay, north Florida, Station 15.
06/87. x1200.

PLATE 41

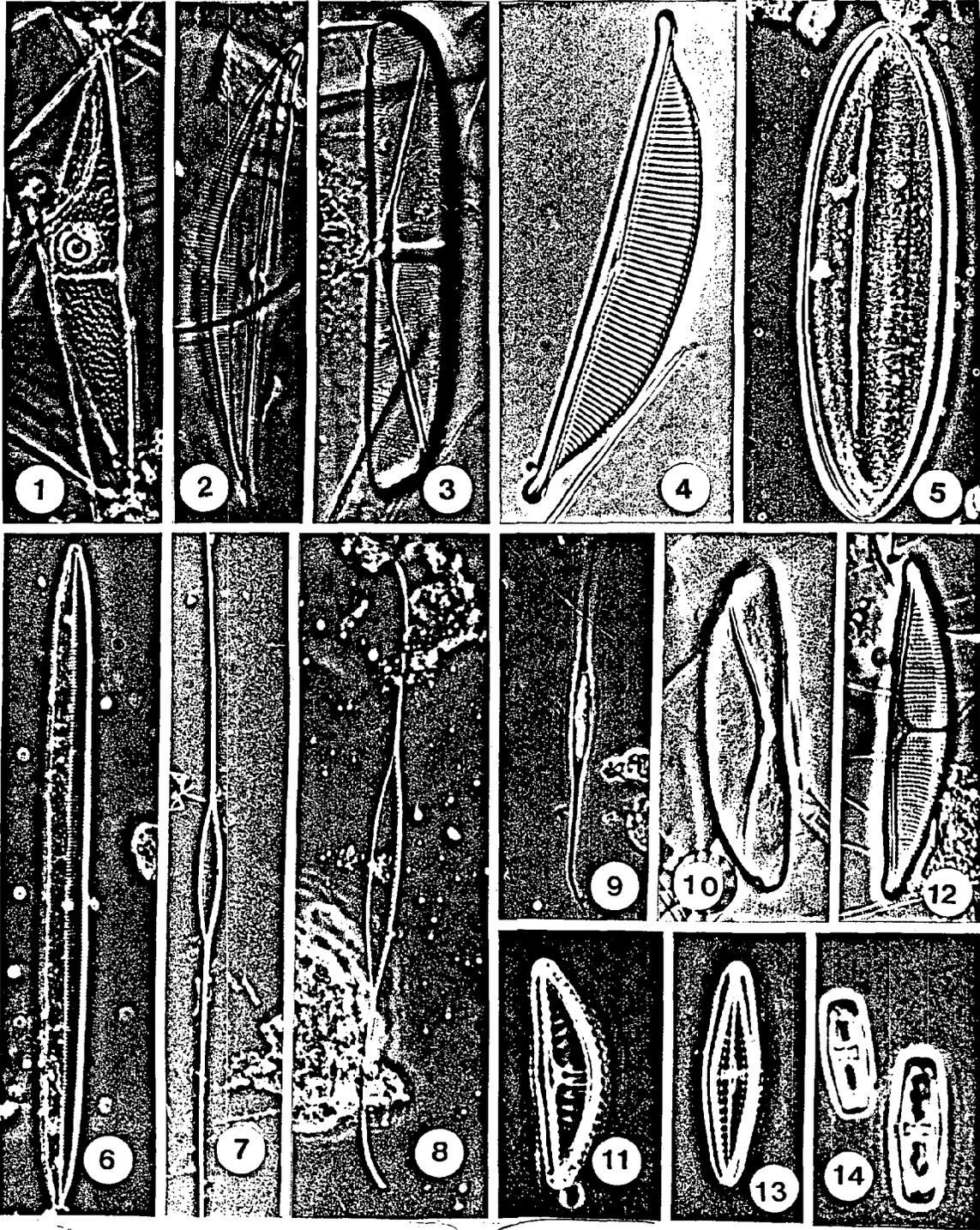


Plate 42

- Fig. 1: *Nitzsca pungens v. atlantica* Cl.
Choctawhatchee Bay, north Florida, Station 36.
02/86. x500.
- Fig. 2: *Nitzsca obtusa* Wm. Sm.
Choctawhatchee Bay, north Florida, Station 03.
01/86. x500.
- Fig. 3: *Nitzsca pellucida* Grunow.
Choctawhatchee Bay, north Florida, Station 25.
02/87. x1000.
- Fig. 4: *Nitzsca lanceolata* Wm. Sm.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 5: *Nitzschia cf. hybrida* Grunow.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 6: *Nitzschia fossilis* Grunow.
Choctawhatchee Bay, north Florida, Station 03.
02/86. x1200.
- Fig. 7: *Nitzschia marginulata* Grun.
Apalachicola Bay, north Florida, Station E-03.
02/88. x300.
- Fig. 8: *Nitzschia epithemoides* Grunow.
Choctawhatchee Bay, north Florida, Station 38.
02/87. x1200.
- Fig. 9: *Nitzschia cf. vitrea* Norman.
Choctawhatchee Bay, north Florida, Station 38.
02/87. x400.
- Fig. 10: *Nitzschia constricta* (Greg.) Grunow.
Choctawhatchee Bay, north Florida, Station 38.
02/87. x400.
- Fig. 11: *Nitzschia reversa* W. Sm.
FSU Marine Lab., north Florida, Station 00.
01/86. x500.

PLATE 42

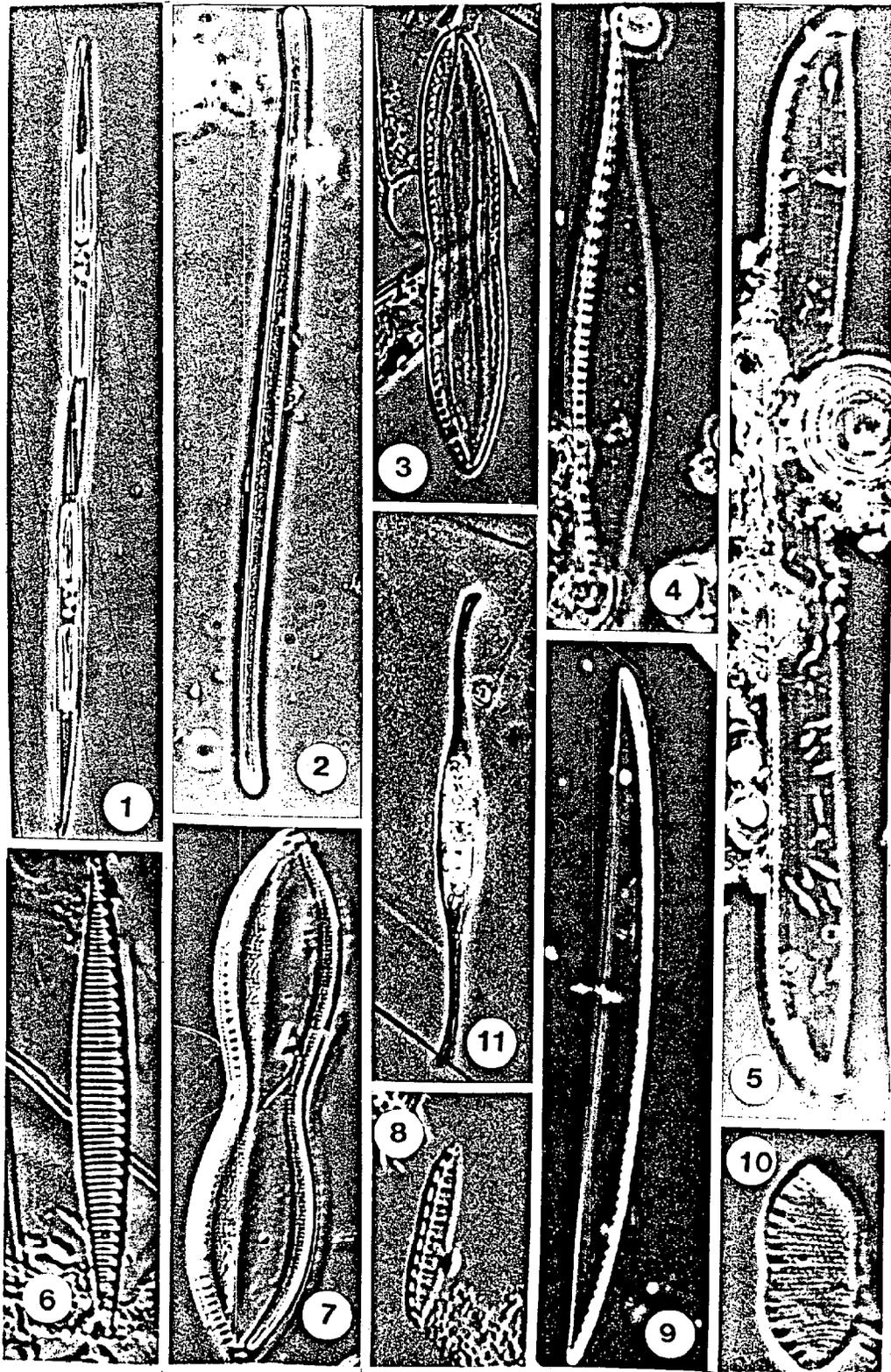


Plate 43

- Fig. 1: *Nitzschia seriata* Cleve.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x250.
- Fig. 2: *Nitzschia Longissima* (Breb.) Ralfs.
Choctawhatchee Bay, north Florida, Station 34.
03/86. x500.
- Fig. 3: *Nitzschia scalaris* (Ehr.) W. Smith.
Choctawhatchee Bay, north Florida, Station 07.
02/86. x250.
- Fig. 4: *Nitzschia sigma* (Kuetz.) W. Sm.
Choctawhatchee Bay, north Florida, Station 07.
02/86. x500.
- Fig. 5: *Campylodiscus echeneis* Ehr.
Choctawhatchee Bay, north Florida, Station 03.
01/86. x500.
- Fig. 6: *Surirella fastuosa* Ehr.
Choctawhatchee Bay, north Florida, Station 03.
01/86. x500.
- Fig. 7: *Nitzschia seriata* Cleve.
Choctawhatchee Bay, north Florida, Station 35.
10/86. x550.
- Figs. 8,9: *Nitzschia fusoides* Ehrlich.
Choctawhatchee Bay, north Florida, Station 15.
03/87. x1200.
- Fig. 10: *Nitzschia pungens* v. *atlantica* Cleve.
Choctawhatchee Bay, north Florida, Station 07.
03/86. x1200.
- Fig. 11: *Surirella robusta* Ehr.
Choctawhatchee Bay, north Florida, Station 03.
03/86. x500.
- Fig. 12: *Campylodiscus clypeus* Ehr.
Choctawhatchee Bay, north Florida, Station C-02.
10/87. x250.
- Fig. 13: *Nitzschia fusoides* Ehrlich.
Choctawhatchee Bay, north Florida, Station 22.
07/86. x1200.

PLATE 43

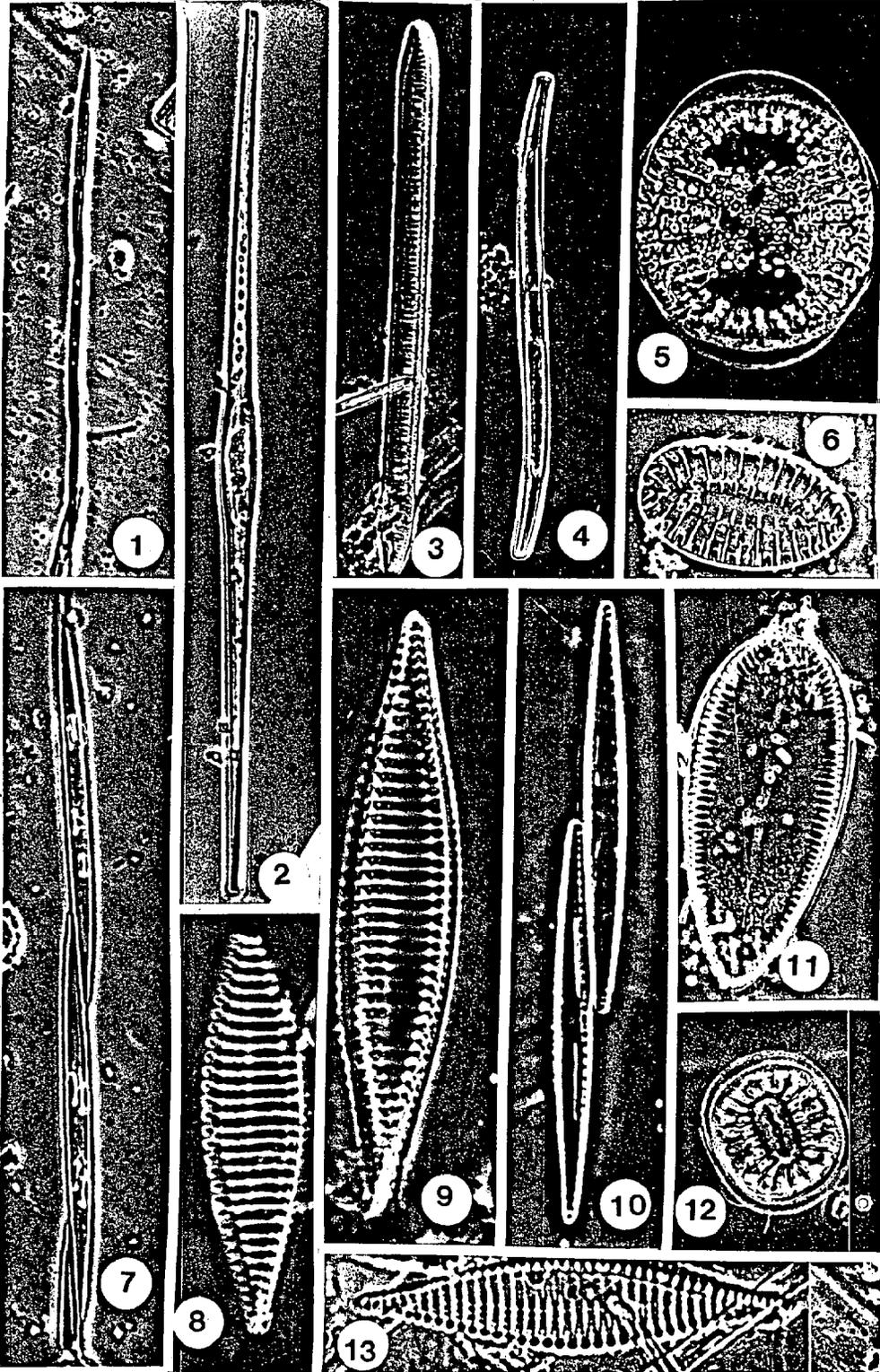


Plate 44

- Fig. 1: *Bacillaria paxillifer* (Mueller) Hendey.
Choctawhatchee Bay, north Florida, station 07.
02/86. x500.
- Fig. 2: *Nitzschia cf. normannii* Grunow.
Choctawhatchee Bay, north Florida, Station 07.
02/86. x1200.
- Figs. 3,4: *Bacillaria paxillifer* (Mueller) Hendey.
Choctawhatchee Bay, north Florida, Station 03.
03/86. Fig. 3: x500. Fig. 4: x1200.
- Fig. 5: *Nitzschia circumscuta* (Bailey) Grunow.
Choctawhatchee Bay, north Florida, Station 15.
03/87. x1200.
- Fig. 6: *Surirella febigeri* Lewis.
Choctawhatchee Bay, north Florida, Station 03.
04/87. x1200.
- Fig. 7: *Nitzschia sigma* (Kuetz.) Wm. Sm.
Choctawhatchee Bay, north Florida, Station 15.
08/87. x1200.
- Fig. 8: *Surirella linearis* W. Sm.
Choctawhatchee Bay, north Florida, Station 07.
01/86. x500.

PLATE 44

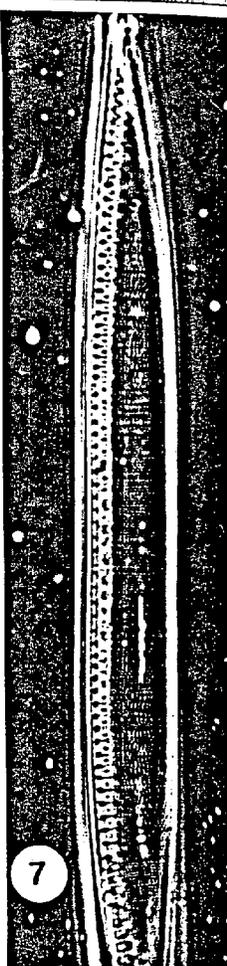
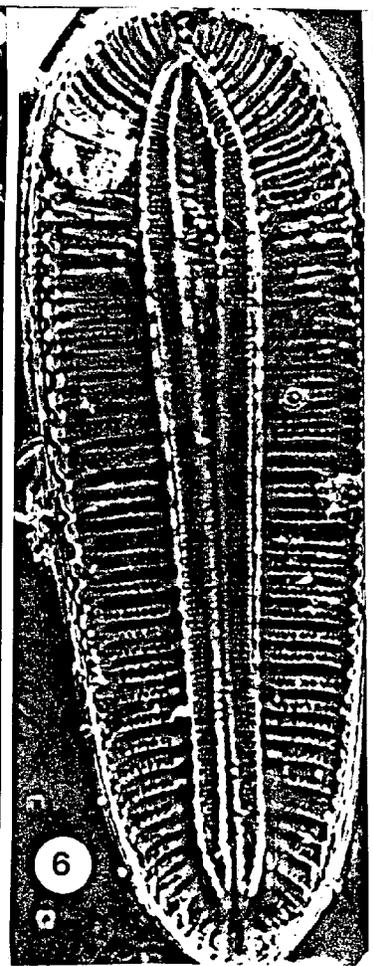
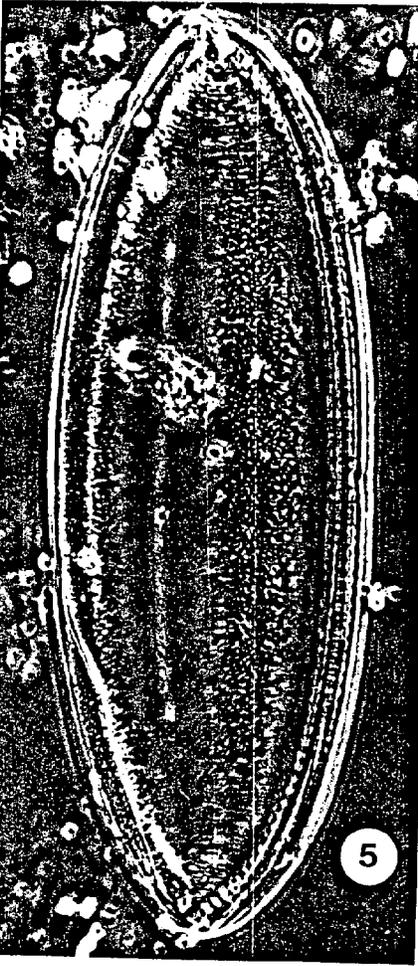
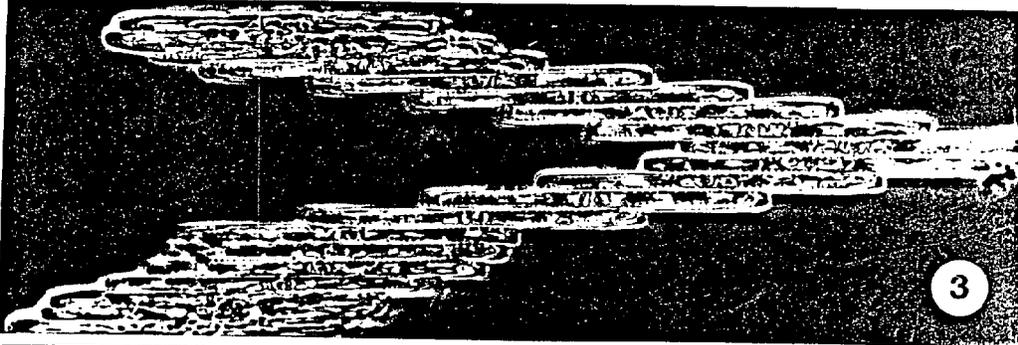
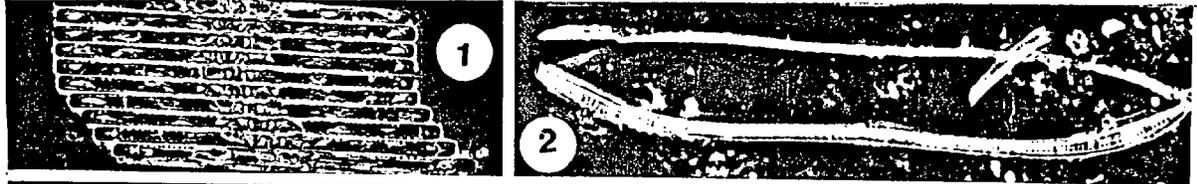


Plate 45

- Figs. 1,2: *Denticula cf. kuetzingii* Grun.
FSU Marine Lab., north Florida, Station 00.
02/86. x1200.
- Figs. 3,4: *Hantzschia* sp.
Apalachicola Bay, north Florida, Station E-01.
02/87. x1200.
- Fig. 5: *Nitzschia sigma* (Kuetz.) Wm. Sm.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 6: *Hantzschia* sp.
Apalcheecola Bay, north Florida, Station E-01.
02/87. x1200.
- Fig. 7: *Denticula cf. kuetzingii* Grun.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 8: *Nitzschia lacunarum* Hustedt.
Choctawhatchee Bay, north Florida, Station C-02.
02/88. x1200.
- Fig. 9. *Nitzschia dubia* Wm. Sm.
Choctawhatchee Bay, north Florida, Station 38.
02/86. x1000.
- Fig. 10. *Bacillaria paxillifer* (Mueller) Hendey.
Choctawhatchee Bay, north Florida, Station 38.
12/86. x1200.
- Fig. 11: *Nitzschia dissipata* (Kuetz.) Grunow.
Apalcheecola Bay, north Florida, Station E-04.
02/88. x1200.

PLATE 45

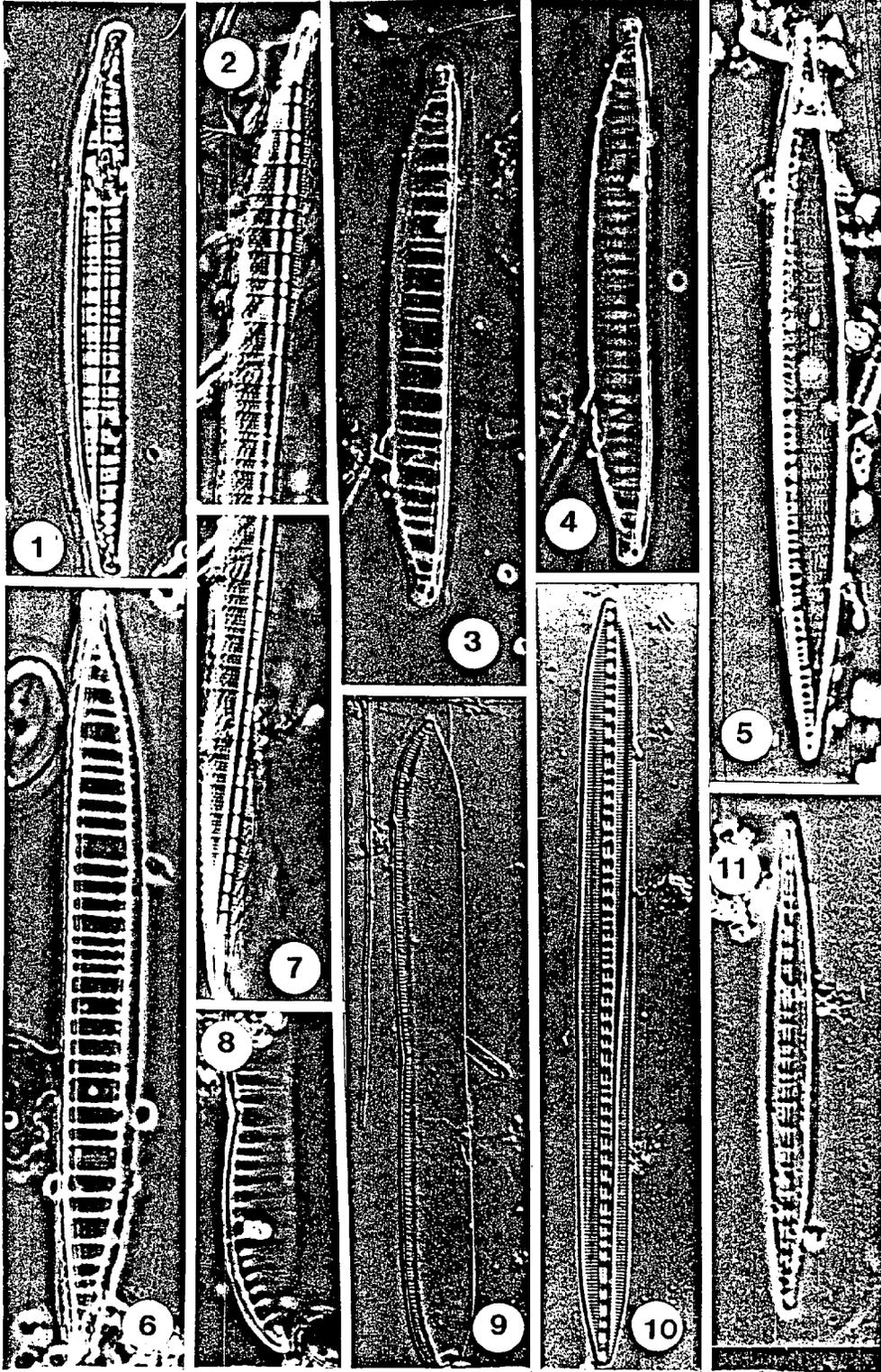
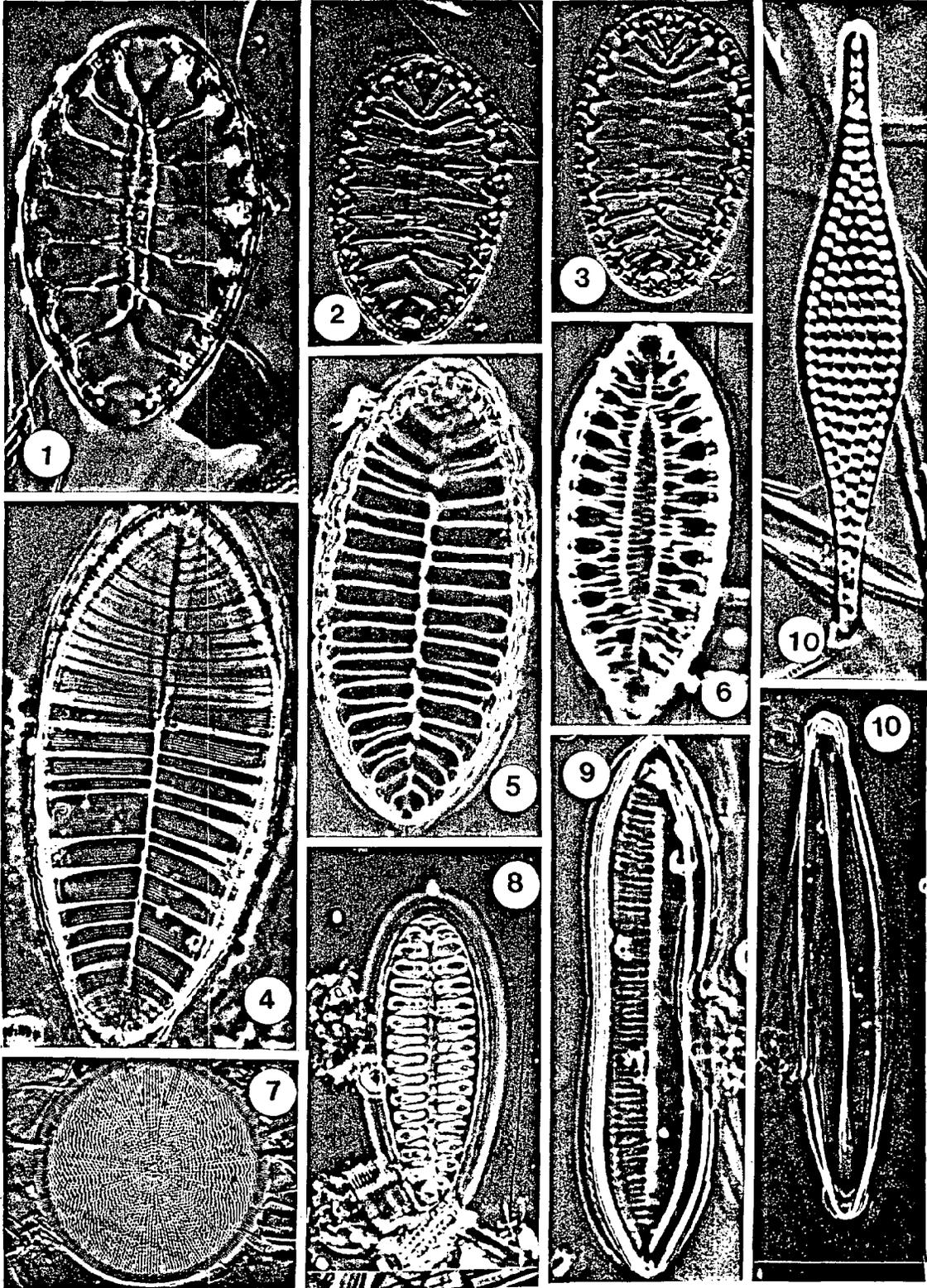


Plate 46

- Fig. 1-3: *Surirella fastuosa* Ehr.
Choctawhatchee Bay, north Florida, Station 38.
01/86. x700.
- Fig. 4: *Surirella gemma* Ehr.
Choctawhatchee Bay, north Florida, Station 03.
04/87. x500.
- Fig. 5: *Surirella linearis* Wm. Sm.
Choctawhatchee Bay, north Florida, Station 03.
04/87. x500.
- Fig. 6: *Surirella amphioxysis* W. Sm.
Apalachicola Bay, north Florida, Station E-04.
02/88. x1200.
- Fig. 7: *Actinocyclus ehrenbergii* Ralfs.
Apalachee Bay, north Florida, Station A-03.
02/88. x500.
- Fig. 8: *Surirella biseriata* Breb.
FSU Marine Lab., north Florida, Station M-03.
10/87. x1200.
- Fig. 9: *Nitzschia levidensis* (W. Sm.) Grun.
FSU Marine Lab., north Florida, Station 00.
01/86. x1200.
- Fig. 10: *Cymatosira lorenziana* Grunow.
Choctawhatchee Bay, north Florida, Station 31.
04/87. x1000.
- Fig. 11: *Striatella unipunctata* (Lyngb.) Ag.
Choctawhatchee Bay, north Florida, Station 31.
12/85. x1200.

PLATE 46



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