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LATE CRETACEOUS DINOFLAGELLATE PALYNOMORPHS IN NORTHERN TEXAS COASTAL PLAIN STRATA

A Thesis

Presented for the

Master of Science

Degree

The University of Tennessee, Knoxville

Christian J. Heine

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Financial assistance was provided by the Discretionary Fund of the University of Tennessee Department of Geological Sciences and Mobil Exploration and Producing Services Inc. I thank Dr. Lee Gibson of Mobil Oil Stratigraphic Laboratory, who arranged for the processing of the thesis samples and for permission to use microscopes in the Mobil Stratigraphic Laboratory.

ABSTRACT

A total of 30 samples were examined from Late Cretaceous strata in northeast Texas, exposed in an arc eight to twenty-four kilometers wide extending from Dallas north-northwest to the Arkansas boarder. Dinoflagellate palynomorphs extracted from these samples yielded a maximum in taxonomic richness of 114 taxa. The majority of the palynomorph specimens are represented by fifty genera, particularly within the families Gonyaulacacae and Peridiniaceae.

The stratigraphic ranges of the dinoflagellate cysts within the northeast Texas section have been established, following the foramanifera zonation and calcareous nannoplankton zonation set up by Thompson (1983) and Percival (in press 1985) respectively.

In earlier palynological studies, Zaitzeff and Cross (1966) dealt with geographically related rocks, whereas Wilson (1974) examined the type Campanian and Maestrichtian sections in Holland, Belgium and Denmark. Identified in this study were twenty-three of the fifty-six dinoflagellate taxa that Zaitzeff and Cross used to zone the Maestrichtian. Furthermore, based on both lithological and paleontological evidence, it has been determined that the Neylandville Marl portion of zone (A) from the Zaitzeff and Cross (1966) study, should be reassigned to the Taylor Group and is possibly Campanian in age.

Comparison with the Wilson (1974) study is limited to zones one and two of Wilson's five dinoflagellate zones, and thirty taxa in those zones were found to be in common with this study. Wilson used the first stratigraphic occurrence of Deflandre diebelii, and Austraiella balmei and the last appearance of Austraiella hvidensis, (Ceratiopsis diebelii, Spinidinium clavum and Spinidinium densispinatum in this study) to identify the Campanian-Maestrichtian boundary in Denmark. In the Northeast Texas section both Ceratiopsis dibelii, and Spinidinium calvum are found in sediments of Campanian age, while the range of Spinidinium densispinatum continues into the Maestrichtian. Wilson uses the last stratigraphic occurrence of Odontochitina operculata, O. costata, Senoniasphaera protrusa and S. rotunda to mark the Campanian-Maestrichtian boundary in the Maestricht region. In Northeast Texas, both Odontochitina operculata and O. Costata extend into the Maestrichtian, while Senoniasphaeia protrusa and S. rotunda end within the Campanian.

Cluster analysis was used to identify five dinoflagellate assemblages considered to be environmentally controlled by depositional environments. Four of these assemblages corresponds to particular lithologies associated with Annona, Gober, Brownstown and Marlbrook formations, respectively. The fifth assemblage is considered to be a transitional assemblage between two depositional environments. Statistical measures of dominance and taxonomic

richness, the Gonyaulacaceae/Peridiniaceae ratio, and percent representation by family, were used in developing the relationships between dinoflagellate taxa and five major depositional environments of the Late Cretaceous marine shelf in northeast Texas.

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CHAPTER I

INTRODUCTION

General Statement

Samples used in this investigation were collected from Upper Cretaceous strata in northeast Texas which crop out in an arc eight to twenty-four kilometers wide extending from Dallas north-northeast to the border with Oklahoma and Arkansas west-northwest of Texarkana (Fig. 1). Continuing southwestward from this area, a diagonal belt of rocks of comparable age crosses central Texas east and south of the Llano Uplift and, from the vicinity of San Antonio, curves westward to the Rio Grande (Fig. 2). Subdivision of these Upper Cretaceous strata into Austinian, Tayloran, and Navarroan stages, correlative with the European standard succession from Coniacian, Santonian, Campanian, to Maestrichtian, remains controversial after nearly a century of study beginning with Hill (1887).

The central Texas section of the Upper Cretaceous outcrop belt in Travis, Williamson, Bell, and McLennan counties has received most attention over the years in attempts to define boundaries, delineate units, and establish reference standards. Fossils from these strata were included in the efforts of Stephenson (1929, 1937) to

Figure 1. Outcrop arc from Dallas to Texarcana showing important towns, roads and sample locations.

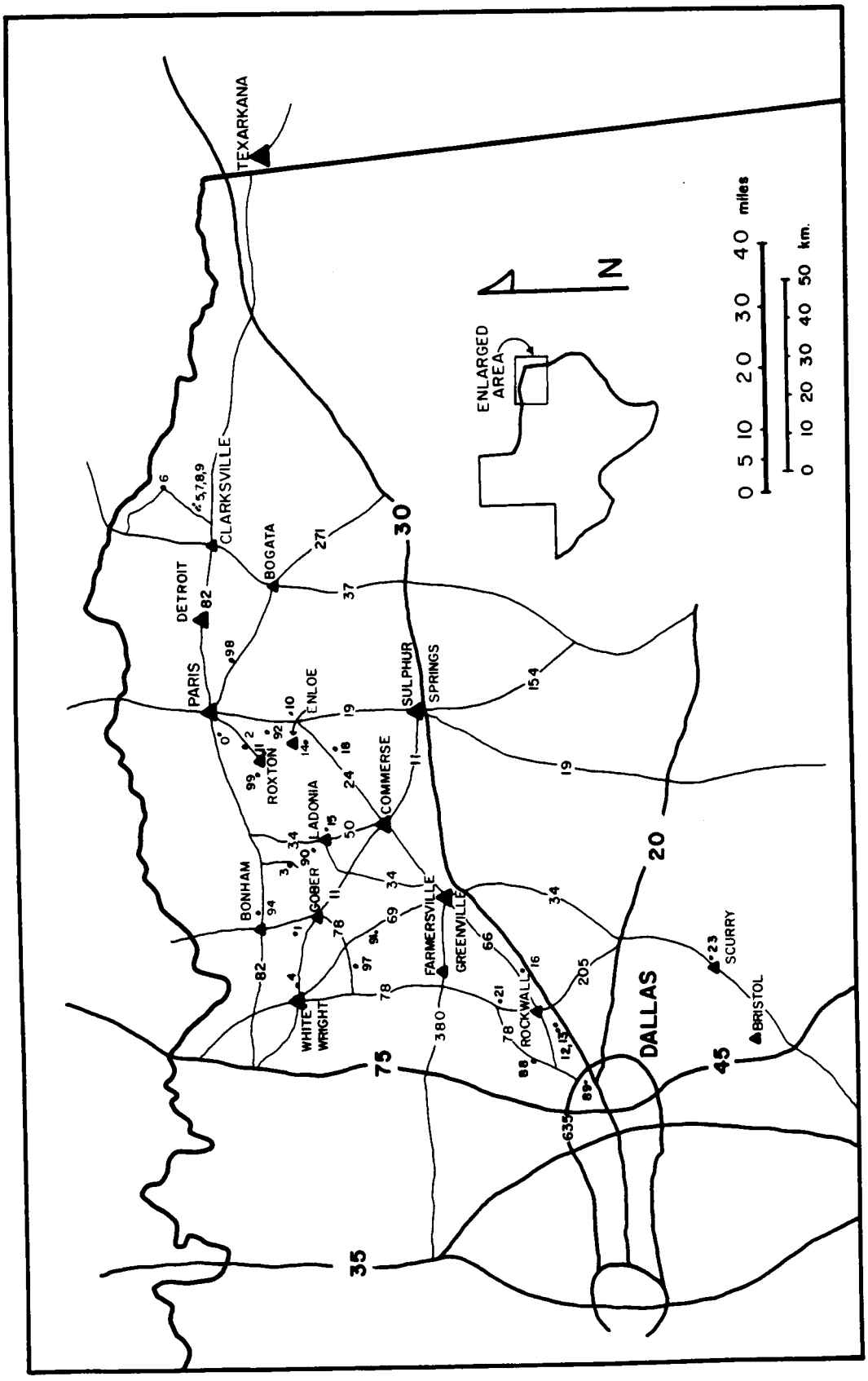

TEXARKANA
CLARKSVILLE
DETROIT
BOGATA
PARIS
ENLOE
SULPHUR SPRINGS
ROXTON
COMMERSE
LADONIA
GOBER
BONHAM
FARMERSVILLE
GREENVILLE
ROCKWALL
DALLAS
SCURRY
BRISTOL
WHITE WRIGHT
ENLARGED AREA
N
0 5 10 20 30 40 miles
0 10 20 30 40 50 km.
30
20
75
35
45
271
82
380
635

Figure 1.

Figure 2. Upper Cretaceous outcrop in Texas.

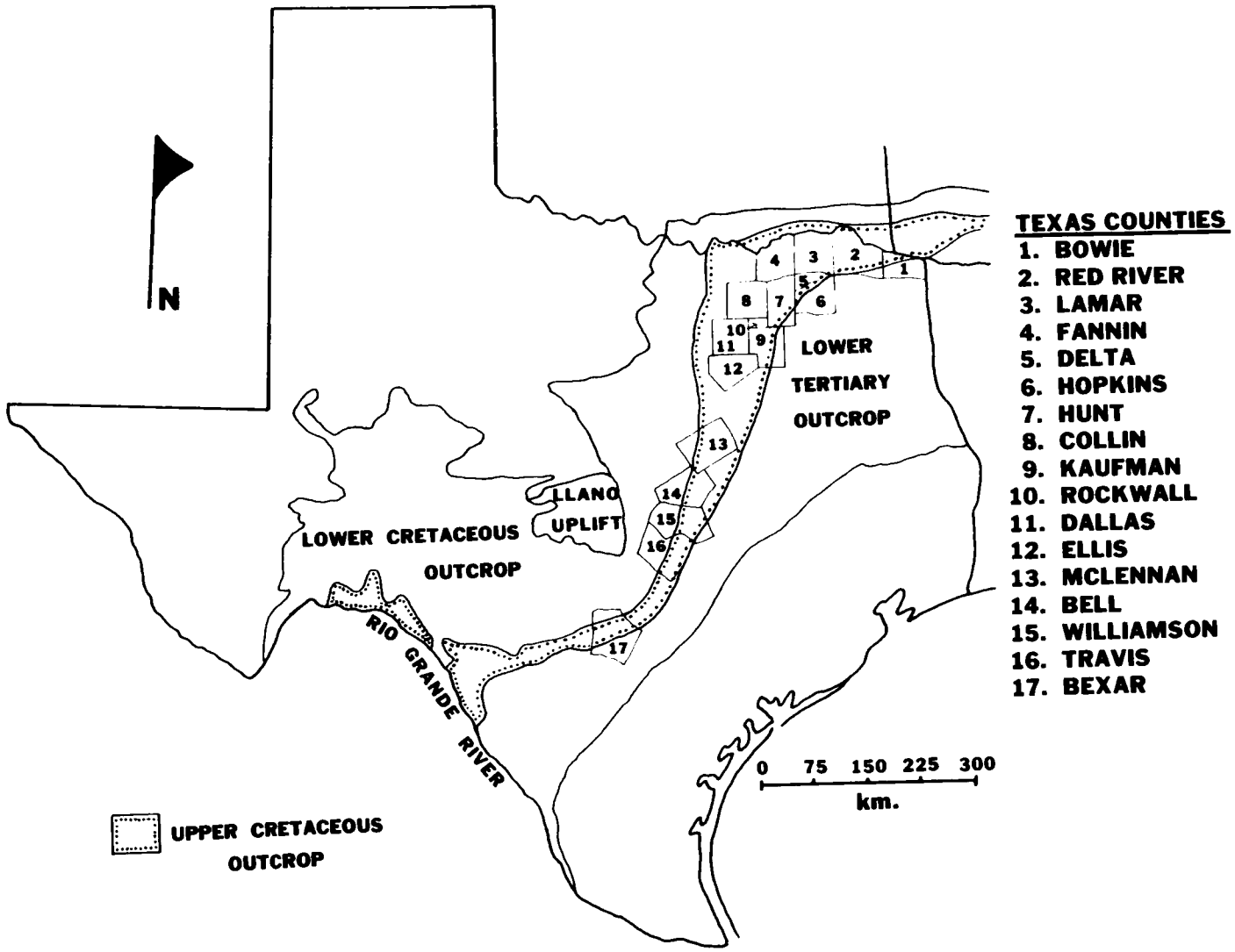

N
LOWER TERTIARY OUTCROP
LLANO UPLIFT
LOWER CRETACEOUS OUTCROP
RIO GRANDE RIVER
UPPER CRETACEOUS OUTCROP
0 75 150 225 300
km.
TEXAS COUNTIES
1. BOWIE
2. RED RIVER
3. LAMAR
4. FANNIN
5. DELTA
6. HOPKINS
7. HUNT
8. COLLIN
9. KAUFMAN
10. ROCKWALL
11. DALLAS
12. ELLIS
13. MCLENNAN
14. BELL
15. WILLIAMSON
16. TRAVIS
17. BEXAR

Figure 2.

zone Late Cretaceous stratigraphy in the Atlantic and Gulf Coastal Province on the basis of the Inoceramus - Gryphaea-Exogyra - Ostrea oyster succession. Later detailed lithostratigraphic studies, notably by Young and Marks (1952) in Williamson County and by Durham (1957) from Bell County to the Rio Grande, challenged formational and stage boundaries of the earlier workers. In northeast Texas, Durham (1957) contended that Austinian clays and marls had been incorrectly assigned to the Tayloran stage. More recently, Thompson (1983) has shown, based on foraminiferal evidence, that many of the formations are time transgressive. Replacing the problematical standard stage divisions with foraminiferal biozones, designated by letters A through F, Thompson placed the Upper and Lower Campanian boundaries at the top of zone E and at the top of zone A respectively. In effect, beds included in both the lower Navarro and the upper Austin North American stages are regarded by Thompson (1983) as being Campanian in age.

For the present study, thirty samples spanning the Upper Santonian through the lower Maestrichtian were selected as a focus for palynological investigation. The samples were collected in the northeastern Texas counties of Fannin, Lamar, Red River, Collin, Hunt, Delta, Rockwall, Dallas and Kaufman. In choosing the samples for processing, care was exercised to select both the best possible vertical

representation within each formation, as well as good lateral distribution throughout the time transgressive units within each of the six biozones set up by Thompson (Fig. 3). The same thirty samples were also processed for calcareous nannoplankton and interpreted by Percival (in press), providing complementary stratigraphic information for these sections. Percival followed the nannoplankton zonation established by Sissingh (1977, 1978). Figure 3 displays the stratigraphic position of all the samples used in this thesis relative to the foraminiferal and calcareous nannoplankton zonations of Thompson and Percival, respectively.

Previous Palynological Studies

Few published reports of palynological studies are available emphasizing dinoflagellates from Upper Cretaceous rocks of the Gulf Coastal Plain. This palynological literature covering the Upper Cretaceous of Northeast Texas comprises: (a) a dissertation by Zaitzeff (1967) representing a taxonomic and stratigraphic study of Maestrichtian-age Navarro group rocks; (b) a report on the zonation of the Navarro group using dinoflagellates and acritarchs (Zaitzeff and Cross 1966) and; (c) an interpretation of the stratigraphic relationships of the Annona Chalk and the Gober Chalk (Campanian), (Patricelli 1978). In addition, relevant and useful palynologic studies involving Cretaceous dinoflagellates within strata of the Atlantic Coastal Plain include studies by May (1976), Whitney (1976), and Benson (1975).

NANNO ZONE	FORAM ZONE	AUSTIN CHALK	BROWNSTOWN MARL	GOBER CHALK	ANNONA CHALK	WOLFE CITY SANDSTONE	PECAN GAP CHALK	MARLBROOK MARL	NACATOCH SANDSTONE
23a	F							18	23
22	E							21 14	
21							15		
20- 19b	D				6 5	11 10	16		
			90						
19a — 18b	C		12 13 91 92	98 0	9 8 7				
18a				97 99					
	B			2					
17		89 88		1					
	A		94	3					
16			95	4					

Figure 3. Distribution of the 30 samples from eight formations, and their relation to the foraminiferal and calcareous nannoplankton zones of Thompson (1983) and Percival (in press 1985) respectively.

Drugg (1967) described Maestrichtian-Paleocene assemblages from California. Hardland (1973) investigated the dinoflagellate cysts and acritarchs from the Bearpaw Formation (Upper Campanian) of both Alberta and Montana. Probably the most studied Upper Cretaceous strata on the North American Continent are those off of southeast Canada in the Scotian Shelf - Grand Banks area. Important papers from this area include Bujak and Williams (1978), Williams and Lentin (1975), Williams (1975), Williams and Brideaux (1975), and Barss, Bujak and Williams (1979).

Gelogic Setting and Stratigraphy

The Campanian Shelf in northeast Texas has been interpreted by Stehli, et al. (1972) as a broad shallow shelf, gently dipping to the southeast. The units comprising this section are composed primarily of marls, with a few thin units of chalk and sandstone. The five lithologic units that make up the Upper Cretaceous strata of northeast Texas are, in stratigraphic order, from youngest to oldest, the Woodbine, Eagleford, Austin, Taylor and Navarro Group rocks. This study concentrates on the upper Austin, Taylor and the lower Navarro groups. The formations comprising the upper Austin Group are the Austin Chalk and the Gober Chalk. The Taylor Group is made up of five formations, the Brownstown Marl, Wolfe City Sandstone, Pecan

Gap Chalk, Annona Chalk and the Marlbrook Marl. Only one formation, the Nacatoch Sandstone, represents the Navarro Group rocks. These formations are shown (Fig. 4), in a lithofacies distribution map after Thompson (1983, p. 57).

Thompson (1983) found the thickness of the units to be problematic. Numerous normal faults downthrown towards the Gulf were recognized in the field, and, although the displacements were less than three meters, Thompson suggested that the sum of the offsets may be considerable. Formation thickness were calculated by Thompson using unit thicknesses estimated from geologic maps and measured sections. Altogether the strata of the upper Austin, Taylor and lower Navarro groups (the focus of the present study) account for 300-450 meters of section in Northeast Texas. The study area crops out in an arc eight to twenty-four kilometers wide from Dallas to Texarkana (Fig. 1) P. 2. In a traverse perpendicular to strike, the strata would dip approximately 6.2 meters per kilometer.

Austin Chalk

The Austin Chalk consists of chalk and calcareous mudstone. Hill (1901) described the Austin in Williamson County in central Texas and formally divided it into a lower, almost pure chalk, and an upper, more marly chalk. Within the study area, the upper Austin Chalk was examined in both Dallas and Collin counties. According to Thompson

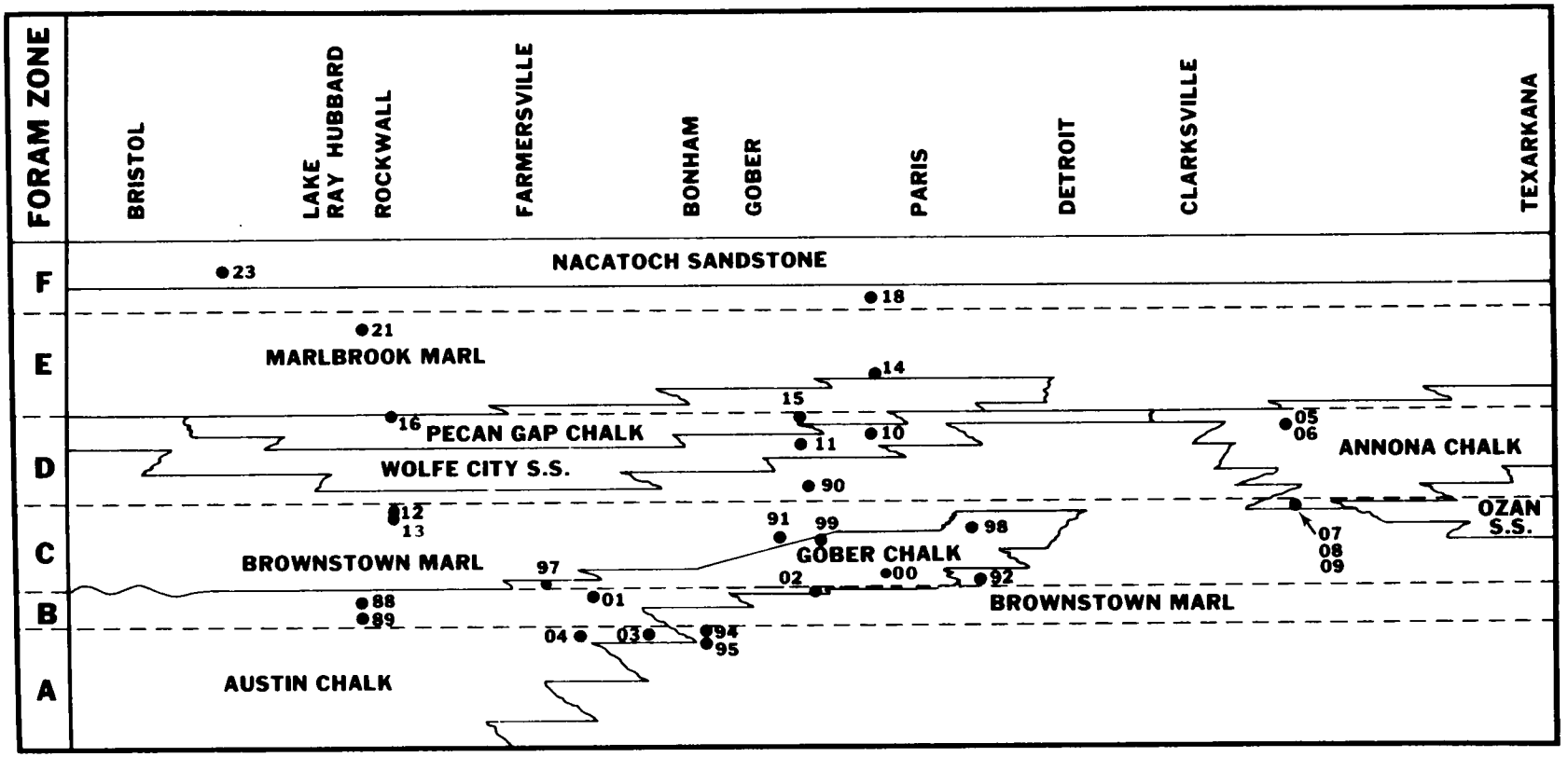


Figure 4. Location map showing distribution of palynomorph samples and lithofacies modified after Thompson (1983, p. 57).

(1983), the Austin Chalk consists of 0.3 - 3.0 m thick chalk beds separated by 10 - 20 cm thick beds of calcareous mudstone. Renalder (1981) described the calcareous chalk as a sparse biomicrite containing whole forams and coccoliths with abundant coccolith debris. Thompson (1983, locality 8-21-6-3) shows the contact of the Austin with the Brownstown Marl as conformable, with the interbedded chalk and calcareous mudstone grading into a uniform calcareous mudstone.

Gober Chalk

The Gober Chalk was first described by Stephenson (1918) as the "Annona Tongue of the Austin Chalk." Stephenson (1927) formally introduced the name Gober for the chalky tongue of the Austin. Stenzel (1938) replaced the name Gober Tongue with the Gober Chalk and assigned the chalk unit to the Taylor group on the basis of fossils. Thompson (1983) pointed out that the assignment of the Gober Chalk to the Taylor group is a lithostratigraphic and not a biostratigraphic problem, and cannot be accomplished by using fossils. Lithostratigraphically, the Gober Chalk is part of the Austin group.

The Gober Chalk is composed of interbedded chalk and calcareous mudstone. In outcrop, the chalks weather to white flaggy units. According to Thompson (1983), the lower contact of the Gober Chalk is in fault contact with the Brownstown Marl and is therefore undefined. However, as

observed by Thompson (1983), the upper contact of the Gober Chalk with the Brownstown Marl appears to be conformable.

Brownstown Marl

The most recent study of the Brownstown Marl was carried out by Beall (1964) in which he described the Brownstown as "a dark brown dominantly montmorillinite clay with varying amounts of silt-sized quartz, calcite fragments, glauconite pellets, phosphate nodules, hematite and finely-disseminated pyrite or pyrite nodules" (Beal, 1964, p. 16). In the study area, Thompson (1983) described the Brownstown in outcrop as gray calcareous mudstone exhibiting conchoidal fracture.

In the literature, the strata referred to in this study as the Brownstown Marl appear under many names. Hill (1894) introduced the name Taylor Marl, which Gordon (1909) divided into an upper and lower marl. Earlier, Brownstown as a unit name was first cited by Hill (1888) for beds overlying the White Cliffs Chalk (Annona Chalk). Veach (1905) correctly recognized the Brownstown as underlying the Annona but included the Ozan Formation as part of the Brownstown in his description. Thompson (1983) formally amended the Brownstown Marl to include the lower Taylor Marl and the Brownstown Marl of other authors.

The contacts of the Brownstown with both the Austin and Gober chalks appears to be conformable. The upper

contact of the Brownstown Marl with the overlying Wolfe City Sandstone also appears conformable (Thompson, 1983).

Wolfe City Sandstone

Richardson (1972) divided the Wolfe City Sandstone into two members: an upper member consisting of 0 - 21 m of very fine-grained, friable to well-cemented sandstone; and a lower member consisting of about 15 m of sandy calcareous clay. McNulty et al. (1981) named the "upper member" the Farmersville Member of the Pecan Gap Chalk. Thompson (1983) determined that the two lithofacies are not superimposed, as suggested by Richardson (1972) and McNulty et al. (1981), but are lateral equivalents.

The lower contact of the Wolfe City Sandstone has been cited by Richardson (1972) as sharp but conformable. In the southernmost edge of the study area, the Wolfe City Sandstone is overlain by the Marlbrook Marl. At the upper contact, the Wolfe City Sandstone is iron-stained and contains well-developed phosphate nodules. For this reason, Thompson (1983) suggests the contact is disconformable. At the type locality of the Pecan Gap Chalk, Thompson (1983) recognized a disconformable, undulatory contact between the Pecan Gap Chalk and the underlying Wolfe City Sandstone, overlain by a 10 cm thick unit containing pelecypod megafossils and phosphate nodules. In its easternmost extent, the Wolfe City Sandstone is again overlain by the Marlbrook Marl

where a precise contact has not been described, Thompson (1983) suggests that the finer grained facies of the Wolfe City Sandstone grades conformably into the Marlbrook Marl.

Pecan Gap Chalk

Several studies of the Pecan Gap Chalk were conducted by students at Southern Methodist University including Steinhoff (1948), Brown (1949), Goldberg (1948), and Clement (1950), and at the University of Texas at Arlington including Brezina (1974) and Maluf (1975). In a synthesis of these studies, McNulty et al (1981) recognized two lithofacies, an argillaceous chalk named the Rockwall Member, and a sandy chalk termed the Lavon Member. The nature of the lower contact of the Pecan Gap Chalk has been debated for years, but according to Thompson (1983, p. 45) "There appears to be some current agreement that it is unconformable from Red River to Collin County, and conformable from there southward." The upper contact of the Pecan Gap Chalk with the Marlbrook Marl has consistently been reported as conformable.

Annona Chalk

Thompson (1983, p. 48) described the well-indurated Annona Chalk as "...massive, slightly argillaceous blue gray chalk which weathers white." He also states that it is locally sandy, and commonly contains small quantities of glauconite. In all cases where the upper and lower contacts of the Annona Chalk were observed, Thompson (1983) found them to be conformable.

Marlbrook Marl

The Marlbrook Marl was first described by Hill (1888, p.72) referring to the unit as the "Marlbrook-Columbus, or Gryphae vesicularis chalks or marls." As originally described, however, Hill included some of the Saratoga Chalk, Nacatoch Sandstone and Arkadelphia Marl. Veach (1905) restricted the name Marlbrook to the marls which overlie the Annona Chalk but continued to include parts of the Saratoga and Nacatoch Formations. Dane (1926) was the first to use the name Marlbrook in placing the marls stratigraphically between the Nacatoch Sandstone and the Pecan Gap and Annona Chalks.

Adkins (1933) formalized the name Neylandville marl, and described it as a dark brownish-gray clay with glauconite, concretionary limestone nodules, and clay-ironstone nodules. According to Stephenson (1918), inclusion of the Exogyra cancellata zone places the Neylandville Marl in the Navarro Group. In contrast, Beall (1964) examined the Neylandville Marl and the "Upper Taylor" Marl and, based on their lithologies, suggested that they were the same and mapped them as one unit belonging to the Taylor Group.

Thompson (1983, p. 56) formally extended use of the name Marlbrook Marl to include the "Upper Taylor" Marl and Neylandville Marl of earlier authors Veach (1905), Gordon (1909), Stephenson (1918), Adkins (1933), Stephenson (1941), Frizzel (1954) and Beall (1964)].

Beall (1964, p. 22) described the Marlbrook lithology (Neylandville Marl) as "light gray, dominantly montmorillonitic clay with varying amounts of calcite fragments, silt-sized quarts, glauconite, and disseminated pyrite". Wherever observed, the upper contact with the Nacatoch Sandstone, according to Thompson (1983), appears to be disconformable.

Methods of Study

Sample Collection and Processing

Samples used in this investigation were originally collected by Dr. Laird B. Thompson, for use in his palentological investigation of the Campanian strata of Northeast Texas. Detailed sample locations can be found in Appendix A. In choosing the samples to be used for this study, care was exercised to select both the thickest stratigraphic sequences within each formation as well as a good lateral distribution within the time transgressive units of the six biozones set up by Thompson (1983). The same samples chosen for this study were also processed for calcareous nannoplankton and interpreted by Percival (in press 1985), to give additional stratigraphic control through the section.

Samples were processed by Clark Geological Services, of Freemont, California. A copy of the processing technique accompanied the prepared slides, and a description of the process can be found in Appendix B. Permission to reproduce

the processing technique was given from both Joyce Clark, of Clark Geological Services, and Dr. William Evitt, Stanford University, where the processing technique was developed (Guide to Sample Preparation, 1981).

Counting Procedure

For the counting procedure followed in this study it was necessary to establish a standard for the minimum number of specimens to be counted for each stratigraphic sample. Wilson (1959), in plotting total species versus the number of specimens encountered, found that in samples with low species dominance and high species richness, a count of 250 specimens was enough to insure good species representation. In the present study, the same plotting procedure showed that no additional species were encountered beyond the count of 250 specimens (Fig. 5). A conservative count of 300 specimens was used in order to ensure adequate representation. In addition to the minimum 300 count per sample, a full scan of each slide was made to search for any rare species possibly missed in the 300 count, and to locate specimens for photographic purposes.

During the counting phase, twenty-seven samples had counts of at least 300, three samples had 54 or fewer dinoflagellates in the count, and six samples of the thirty-six samples were found to be barren. When possible additional slides were prepared from leftover residues of

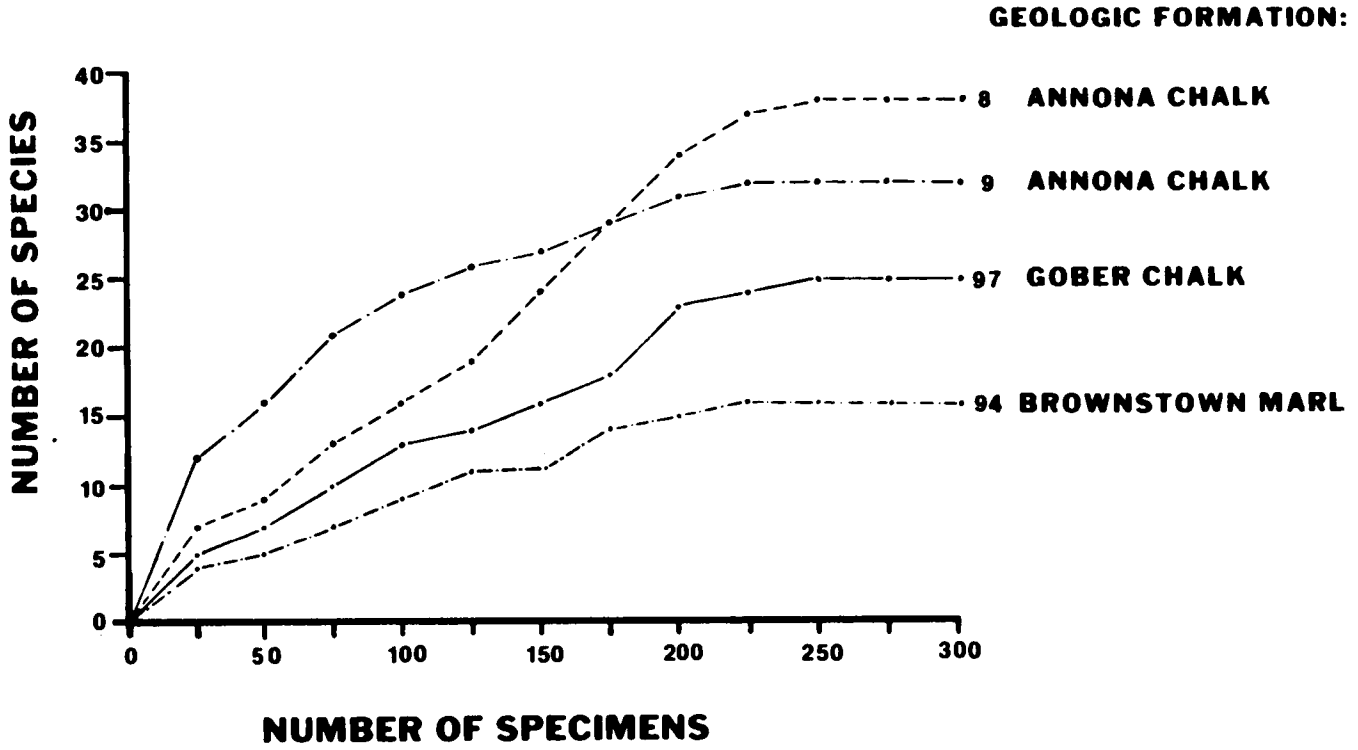


Figure 5. Taxonomic richness (species diversity in the sense of Goodman 1979) versus specimen abundance in Samples 8, 9, 94 and 97.

the barren samples. A second scan of the material again showed the six samples to be barren.

Location of the Specimens.

Slides are stored in the paleontological collections of the University of Tennessee, with accession numbers placed on the slide labels along with the slide number used in this thesis. Figured specimens on a given slide can be located by using the horizontal and vertical stage distances from the center of a scribed cross (0,0 reference point) on the slide near the lower left-hand corner of the cover slip (Appendix C). Coordinates expressed in mechanical stage divisions (millimeters) to the right (horizontal) and above (vertical) of the scribed reference point are given with each figured specimen in the thesis.

Counting was accomplished using a Leitz Wetzlar microscope, University of Tennessee number 130449. All slides were counted at 400X magnification.

Photography

The specimens photographed and figured in this thesis (Appendix C) were photographed with NPL Fluotar objectives using a Leitz Dialux microscope and with a Wild Photoautomat MPS 45 with an automatic metering system. Most of the specimens were photographed at 400X, while some of the larger specimens were photographed at 200X. The ASA of the film (Kodak Tec-Pan ASA 25) was preset by the operator, and proper

exposures were adjusted by varying the exposure time. The Wild Photoautomat MPS 45 utilized a spot metering system which allowed precise metering on the palynomoph of interest. Several exposures of each specimen were taken to ensure a useable photograph. Since the palynomorph color (darkness) varies from specimen to specimen, metering was done on a medium colored specimen, setting the exposure time to give a correct exposure. A series of three pictures were taken for each cyst, one at the correctly metered exposure, one at 1/2 times the exposure time and one at 2 times the exposure time. The background surrounding the palynomorph remained fairly consistent from photograph to photograph. Extremely light or dark specimens would have to be compensated for by varying the exposure times shorter or longer. This technique is similar to using the grey card when doing black and white (or color) photography.

The film was processed using the special developer formulated by Kodak for Tec-Pan film called Technidol. Prints were made on Kodak Polyfiber F single weight paper, which were trimmed and dry mounted for final plates.

CHAPTER 2

RESULTS OF PALYNOLOGICAL INVESTIGATION

Introduction

One hundred and fourteen (114) species belonging to fifty dinoflagellate genera were recognized among the palynomorphs isolated by sample processing. An additional ten taxa have been figured but not identified to genus or species. Pollen and spores were seen on a few slides but no attempt was made during this study to study their taxonomy or stratigraphic distribution. Pollen and spore grains with sizes smaller than 34 microns were inadvertently eliminated by sieving during the initial processing technique. Other organic remains encountered during the total slide scan include acritarchs, microforaminifera linings, scolecodonts, plant fragments and amorphous organic material.

Stratigraphic Palynology

The stratigraphic distribution of each dinoflagellate species is presented in two ways. (1) The abundance for each species is given for every sample. The arrangement of the samples follows the stratigraphic sequence provided by the planktonic foraminiferal and calcareous nannoplankton zonation of Thompson (1983) and Percival (in press 1985), respectively (Table 1). (2) A local range chart, illustrates the dinoflagellate species organized according

Table 1. Numerical distribution of dinoflagellate cysts within the six foraminiferal biozones.

Foram Biozone	Number of Species	Number of First Stratigraphic Occurrances	Number of First Stratigraphic Occurrances	Number of Species Restricted to the Biozone
F	49	8	*	*
E	34	2	7	5
D	61	2	16	8
C	88	26	15	9
B	48	14	1	3
A	44	*	*	*

* Value cannot be determined with available data.

to their first stratigraphic occurrance within the same composite section (Fig. 6).

Appendix D includes a complete species list citing authors and literature references arranged in the order of appearance in Table 1.

A numerical distribution of the 114 dinoflagellate species throughout the six foraminiferal biozones of Thompson (1983) can be seen in Figure 7.

Figure 6. Dinoflagellate count of 300 arranged according to the first stratigraphic occurrence. Stratigraphic position of the samples follows the foraminiferal and calcareous nannoplankton zonation of Thompson (1983) and Percival (in press 1985), respectively.

SAN-TONIAN	CAMPANIAN																MA	AGE
G	B	G	A	G	B	G	An	B	An	W	PG	W An	PG	M		N		FORMATION NAME
16	17			18a	18b		-	19a	19b			20-	21	22		23a		NANNO-PLANKTON ZONE (PERCIVAL, IN PRESS, 1985)
A			B		C				D				E			F		FORAM ZONE (THOMPSON, 1983)
42	322	301	302	7	303	312	300	326	305	300	306	302	310	305	300	310	301	303	306	300	310	300	54	307	308	312	304		TOTAL SPECIMENS
10	20	16	28	2	17	28	29	24	28	9	23	27	33	32	26	40	16	30	38	21	20	23	4	10	12	22	27	33		TOTAL SPECIES
4	95	94	3	88	89	1	2	99	97	92	91	0	98	9	7	8	13	90	6	10	16	5	11	15	14	21	18	23		SAMPLE NUMBER
																														1. Unknown sp. A.
1	5							4			6			1																2. Subtilisphaera sp.
2	20	27	2			+	1		25	2		2	7		4	1		5	7	+	6			1		10				3. Cleistosphaeridium sp.
1	7		18			1	2	6	2			7	18			15		9		2	7			13		12				4. Exochosphaeridium bifidum
1	12					8	21	2						10		4				1							2			5. Canningia scabrosa
+		8	18			4	+	3					35	4		5												5		6. Florentinia clavigera
1		5	2			5	2					7	3	6		2												8	10	7. Chatangiella victoriensis
		+																												8. Chatangiella granulifera
																														9. Palaeohystrichophora infusorioides
																														10. Palaeohystrichophora sp. B.
																														11. Lejeunecysta sp. A.
																														12. Spinidinium echinoideum
																														13. Spinidinium essoi
																														14. Odontochitina porifera
																														15. Coronifera oceanica
																														16. Xenascus ceratioides
																														17. Cyclonephelium distinctum
																														18. Polysphaeridium subtile
																														19. *Trigonopyxidia ginella*
																														20. Dinogymnium acuminatum
																														21. Spiniferities ramosus
																														22. Odontochitina operculata
																														23. Cribroperidinium edwardsii
																														24. Phelodinium tricuspis
																														25. Isabelidinium microarmum
																														26. Apteodinium cribrosum
																														27. Senoniasphaera rotundata
																														28. Spiniferities granulatus
																														29. Hystrichokolpoma rigaudae
																														30. Rottnestia borussica
																														31. Unknown sp B.
																														32. Spiniferities cingulatus
																														33. Trithyrodinium sp A.
																														34. Alterbia acutula
																														35. Unknown sp. C.
																														36. Florentinia sp. A
																														37. Florentinia laciniata
																														38. Senoniasphaera protrusa
																														39. Operculodinium sp. A.
																														40. Fromea sp.
																														41. Exochosphaeridium phragmities
																														42. Hystrichosphaeridium palmatum
																														43. Spinidinium clavus
																														44. Dinogymnium euclaense
																														45. Heterosphaeridium conjunctum
																														46. Isabelladinium acuminatum
																														47. Trithyrodinium sp. B.
																														48. Ceratiopsis pannucea
																														49. Ceratiopsis diebelii
																														50. Laciniadinium arcticum
																														51. Surculosphaeridium longifurcatum
																														52. Diphyes sp. A.
																														53. Florentinia sp. B.
																														54. Canningia sp. A.
																														55. Hystrichosphaeridium tubiferum
																														56. Paleocystodinium Sp. C
																														57. Hystrichosphaeridium sp. X.
																														58. Areoligera senonensis
																														59. Cyclonephelium sp. D.
																														60. Odontochitina costata
																														61. Hystrichosphaeridium sp. A.
																														62. Glaphyrocysta intricata
																														63. Chatangiella tripartita
																														64. Tanyosphaeridium xanthiopyxides
																														65. Nematosphaeropsis pusulosa
																														66. Hystrichosphaeridium tubiferum subsp, brevispinum
																														67. Cyclonephelium compactum
																														68. Palaeoperidinium sp. A.
																														69. Isabelidinium belfastense
																														70. Eatonicysta sp. A.
																														71. Ginginodinium sp. A.
																														72. Hystrichosphaeridium sp. 512
																														73. Impagidinium sp A.
																														74. Alterbia sp. A.
																														75. Dinogymnium sp. A.
																														76. Spinidinium sverdrupianum
																														77. Trithyrodinium sp. C.
																														78. Dinogymnium longicornis
																														79. Unknown X (Ellips spon)
																														80. Unknown Peridinioid 518
																														81. Palaeocystodinium sp A
																														82. Alterbia sp B
																														83. Aiora sp
																														84. Dinogymnium digitus
																														85. Cordosphaeridium fibrospinosum
																														86. Dinogymnium cerviculum
																														87. Spinidinium sp. B.
																														88. Palaeocystodinium benjaminii
																														89. Lejeunecysta sp. B.
																														90. Palaeohystrichophora sp. C.
																														91. Adnatosphaeridium vittatum
																														92. Spinidinium densispinatum
																														93. Dinogymnium sp. B
																														94. Unknown sp. 506
																														95. Deflandre sp. C.
																														96. Ascodinium acrophorum
																														97. Unknown sp 516
																														98. Dinogymnium denticulatum
																														99. Oligosphaeridium sp.
																														100. Dinogymnium sp C
																														101. Unknown sp. G.
																														102. Alterbia sp. C.
																														103. Dinogymnium sibiricum
																														104. Cyclonephelium sp. C.
																														105. Odontochitinopsis sp. A.
																														106. Unknown Peridinioid
																														107. Isabeladinium sp. A.
																														108. Deflandre sp. A
																														109. Deflandre sp B
																														110. Unknown sp. Z.
																														111. Unknown Gonyaulacoid
																														112. Unknown Deflandre
																														113. Spinidinium sp. G.
																														114. Unknown sp E

LEGEND

N-NACATOCH SANDSTONE
M-MARLBROOK MARL
AN-ANNONA CHALK
PG-PECAN GAP CHALK
W-WOLFE CITY SANDSTONE
G-GOBER CHALK
B-BROWNSTOWN MARL
A-AUSTIN CHALK

Figure 25. 6

Figure 7. Dinoflagellate distribution arranged according to first occurrence. Stratigraphic position of the samples follows the foraminiferal and calcareous nannoplankton zonation of Thompson (1983) and Percival (in press 1985) respectively.

SAN-TONIAN	CAMPANIAN																						MA	AGE
G	B	G	A	G		B	G	An	B	An	W	PG	An	W	PG	M		N	FORMATION NAME	
16		17		18a		18b		-	19a	19b		20-			21	22		23a	NANNO-PLANKTON ZONE (PERCIVAL, IN PRESS, 1985)	
A			B			C						D				E		F	FORAM ZONE (THOMPSON, 1983)	
42	322	301	302	7	303	312	300	300	326	305	300	306	302	300	310	305	300	301	303	306	300	300	310	300	54	307	308	312	304	TOTAL SPECIMENS
10	20	16	28	2	17	28	29	24	28	9	23	27	33	32	40	26	24	16	30	38	21	20	23	4	10	12	22	27	33	TOTAL SPECIES
4	95	94	3	88	89	1	2	99	97	92	91	0	98	9	8	7	12	13	90	6	10	16	5	11	15	14	21	18	23	SAMPLE NUMBER

1. Unknown sp. A.
2. Subtilisphaera sp.
3. Cleistosphaeridium sp.
4. Exochosphaeridium bifidum
5. Canningia scabrosa
6. Florentinia clavigera
7. Chatangiella victoriensis
8. Chatangiella granulifera
9. Palaeohystrichophora infusorioides
10. Palaeohystrichophora sp. B.
11. Lejeunecysta sp. A.
12. Spinidinium echinoideum
13. Spinidinium essoi
14. Odontochitina porifera
15. Coronifera oceanica
16. Xenascus ceratioides
17. Cyclonephelium distinctum
18. Polysphaeridium subtile
19. Trigonopyxidia ginella
20. Dinogymnium acuminatum
21. Spiniferities ramosus
22. Odontochitina operculata
23. Cribroperidinium edwardsii
24. Phelodinium tricuspis
25. Isabelidinium microarmum
26. Apteodinium cribrosum
27. Senoniasphaera rotundata
28. Spiniferities granulatus
29. Hystrichokolpoma rigaudae
30. Rottnestia borussica
31. Unknown sp B.
32. Spiniferities cingulatus
33. Trithyrodinium sp A.
34. Alterbia acutula
35. Unknown sp. C.
36. Florentinia sp. A
37. Florentinia laciniata
38. Senoniasphaera protrusa
39. Operculodinium sp. A.
40. Fromea sp.
41. Exochosphaeridium phragmities
42. Hystrichosphaeridium palmatum
43. Spinidinium clavus
44. Dinogymnium euclaense
45. Heterosphaeridium conjunctum
46. Isabelladinium acuminatum
47. Trithyrodinium sp. B.
48. Ceratiopsis pannucea
49. Ceratiopsis diebelii
50. Laciniadinium arcticum
51. Surculosphaeridium longifurcatum
52. Diphyes sp. A.
53. Florentinia sp. B.
54. Canningia sp. A.
55. Hystrichosphaeridium tubiferum
56. Paleocystodinium sp. C
57. Hystrichosphaeridium sp. X.
58. Areoligera senonensis
59. Cyclonephelium sp. D.
60. Odontochitina costata
61. Hystrichosphaeridium sp. A.
62. Glaphyrocysta intricata
63. Chatangiella tripartita
64. Tanyosphaeridium xanthiopyxides
65. Nematosphaeropsis pusulosa
66. Hystrichosphaeridium tubiferum subsp, brevispinum
67. Cyclonephelium compactum
68. Palaeoperidinium sp. A.
69. Isabelidinium belfastense
70. Eatonicysta sp. A.
71. Ginginodinium sp. A.
72. Hystrichosphaeridium sp. 512
73. Impagidinium sp A.
74. Alterbia sp. A.
75. Dinogymnium sp. A.
76. Spinidinium sverdrupianum
77. Trithyrodinium sp. C.
78. Dinogymnium longicornis
79. Unknown X (Ellips spon)
80. Unknown Peridinioid 518
81. Palaeocystodinium sp A
82. Alterbia sp B
83. Aiora sp
84. Dinogymnium digitus
85. Cordosphaeridium fibrospinosum
86. Dinogymnium cerviculum
87. Spinidinium sp. B.
88. Palaeocystodinium benjaminii
89. Lejeunecysta sp. B.
90. Palaeohystrichophora sp. C.
91. Adnatosphaeridium vittatum
92. Spinidinium densispinatum
93. Dinogymnium sp. B
94. Unknown sp. 506
95. Deflandre sp. C.
96. Ascodinium acrophorum
97. Unknown sp 516
98. Dinogymnium denticulatum
99. Oligosphaeridium sp.
100. Dinogymnium sp C
101. Unknown sp. G.
102. Alterbia sp. C.
103. Dinogymnium sibiricum
104. Cyclonephelium sp. C.
105. Odontochitinopsis sp. A.
106. Unknown Peridinioid
107. Isabeladinium sp. A.
108. Deflandre sp. A
109. Deflandre sp B
110. Unknown sp. Z.
111. Unknown Gonyaulacoid
112. Unknown Deflandre
113. Spinidinium sp. G.
114. Unknown sp E

LEGEND

N-NACATOCH SANDSTONE
M-MARLBROOK MARL
AN-ANNONA CHALK
PG-PECAN GAP CHALK
W-WOLFE CITY SANDSTONE
G-GOBER CHALK
B-BROWNSTOWN MARL
A-AUSTIN CHALK

Figure 7
27

CHAPTER 3

INTERPRETATION OF RESULTS

Biostratigraphic Inferences

The inferred stratigraphic position of the samples follows the foraminiferal zonation set up by Thompson (1983), and the calcareous nannoplankton zonation of Percival (in press). Because the sampling represents a composite section, no assemblage or range zones have been proposed, and any informal zonation will be made in reference to Thompson's foraminiferal zones. A comparison of the dinoflagellate distribution from this study is made with the dinoflagellate zonation of the Navarro Group rocks by Zaitzeff and Cross (1966), and the zonation of the Campanian and Maestrichtian of Holland and Denmark by Wilson (1974).

In the palynological study of Navarro Group sediments, Zaitzeff and Cross (1966) utilized two sections, of which only the Austin section included samples of Campanian age. Of the 56 dinoflagellate taxa Zaitzeff and Cross used to zone the Maestrichtian, 23 taxa were common to the list of taxa of the present study. Table 2 lists the species shared by both studies. The taxa of the Zaitzeff and Cross study have been amended where necessary to conform with taxonomic changes made since 1966. Zaitzeff (1967) divided the Navarro Group rocks of Texas into four formations, Neylandville, Marl, Nacatoch Sandstone, Corsicana Marl

Table 2. Taxa observed in both Zaitzeff and Cross (1966) and this study.

Zaitzeff and Cross (1966)	This study
Hystrichosphaeridium sp.1	*Hystrichosphaeridium* sp.A
Paleostomocystis sp.1	*Fromea* sp.
Tanyosphaeridium sp.1	*Tanyosphaeridium xanthiophyxides*
Forma F. sp.1	*Coronifera oceanica*
Cordosphaeridium fibrospinosum	*Cordosphaeridium fibrospinosum*
Hystrichosphaera ramosa var.1.	*Spiniferities granulifera*
H. ramosa	*S. ramosus*
Cyclonephelium sp.1	*Heterosphaeridium conjunctum*
Forma B sp.1	*Senoniasphaera protrusa*
Tenua sp.2	*Cyclonephelium distinctum*
Areoligera senonensis	*Areoligera senonensis*
Dinogymnium sp.1	*Dinogymnium euclaense*
Dinogymnium digitus	*Dinogymnium digitum*
Dinogymnium sp.4	*Dinogymnium denticulatum*
Diconodinium sp.1	*Laciniadinium arcticum*
Gonyaulax sp. 1	Unknown Gonyaulacoid
Odontochitina striatoperforata	*Odontochitina costata*
Forma c. sp.2	*Xenascus ceratioides*
Delflandrea acuminata	*Isabelidinium acuminatum*
D. sp.6	*Isabelidinium microarmum*
D. pannucea	*Ceratiopsis pannucea*
Palaeohystrichophora sp.1	*Palaeohystrichophora* sp.B.
P. infusorioides	*P. infusorioides*

and Kemp Clay, given in order of oldest to youngest, and corresponding to the stratigraphy given on a 1937 U.S.G.S. geological map of Texas. Zaitzeff and Cross (1966) followed the views of Stephenson (1937) that (1) the Exogyra costata faunal zone extended from New Jersey to Mexico and is "co-extensive" with the strata of the Navarro Group, and (2) the Exogyra cancellata zone is restricted to the base of the E. costata zone and thus marks both the base of the Navarro Group sediments (Neylandville Marl), and the base of the Maestrichtian stage. However, Beall (1964) studied the Upper Taylor Marl and the Neylandville Marl and, recognizing no significant lithological difference, considered them both Upper Taylor Marl. More recently, Thompson (1983) has formally proposed the name Marlbrook Marl to include the Upper Taylor Marl, Neylandville Marl and Marlbrook Marl of previous authors.

On biostratigraphic grounds, Frizzel (1954. Fig. 1) placed the Neylandville Marl into the Taylor Group based on foraminifera similarities. Frizzel showed the Neylandville foraminiferal assemblage had 41 species common to both the Taylor Marl and the Navarro Group, 38 species in common with the Taylor Marl only, and 9 species in common with the Navarro Group alone, Zaitzeff (1967). Analysis of a dinoflagellate assemblage from the Neylandville-Nacatoch zone A, Austin section, supported Frizzel's findings. Although the Taylor Mark phytoplankton were beyond the scope

of his study, Zaitzeff recognized several species occurring in the Neylandville Marl, whose ranges did not extend stratigraphically higher than the Neylandville Marl, and are recorded in rocks of the same or older ages from other areas. Zaitzeff (1967 p.131) concluded that the study of the Neylandville microplankton shows that there is a closer relationship of the zone A assemblage to the Taylor Group than to the Navarro. Figure 8 shows the dinoflagellate zonation of Navarro Group rocks by Zaitzeff and Cross (1966). Thus both lithologic and fossil evidence, by Beal (1964), Zaitzeff (1967), and Frizzel (1954) support the assignment of the Neylandville (Marlbrook) Marl to the Taylor Group.

Zaitzeff (1967) does not explain the basis upon which he placed the Maestrichtian stage boundary at the Navarro - "Taylor" contact. If a stage boundary coincident with the Navarro contact is only an assumption, there is a possibility that part of zone A studied by Zaitzeff is actually Campanian.

Wilson (1974) proposed an informal biozonation using the stratigraphic ranges of certain dinoflagellate species. The zones set up by Wilson were classified as concurrent range zones and utilized the overlapping ranges of several species. Wilson (1974) was able to recognize five well defined zones within the Maastricht Region and Denmark.

The Campanian-Maastrichtian boundary in the Maastricht Region correlates with the top of Wilson's zone one (1b).

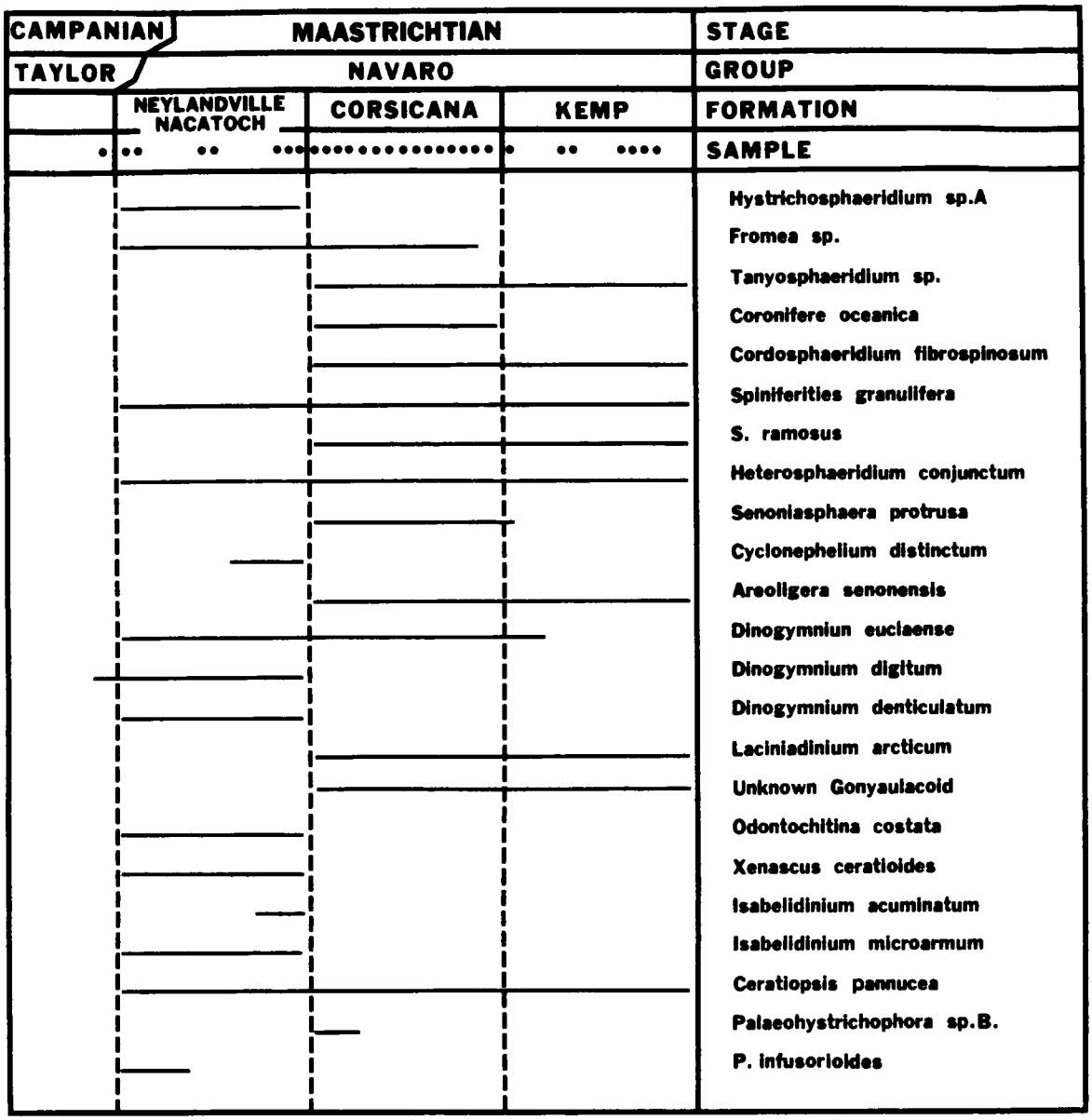


Figure 8. Stratigraphic ranges of the 23 dinoflagellate taxa common to Zaitzeff and Cross (1966) and this study.

In the Denmark section the Campanian-Maastrichtian boundary falls in the middle of Wilson's zone two.

The Northeast Texas samples examined (this study) have thirty taxa in common with the Wilson study. Figure 9 shows the Wilson (1974) zonation using the 30 samples in common with this study. Table 3 shows a listing of the comparable species from Wilson (1974) and this thesis.

Wilson (1974) felt that although the Maastricht Region and Denmark were 600 km apart, a composite zonation of the two sections would be more useful than two separate zonations.

Zone one, the oldest zone recognized by Wilson (1974) was found only in the Maastricht Region and corresponded to the mucronata belemnite zone of Schmidt (1959), and the foraminiferal zone A of Hofker (1966). Zone two in both the Maastricht Region and Denmark corresponds to the langei and lanceolata belemnite zone and is equivalent to the foraminiferal zone B of Hofker (1966).

Zones three through five of Wilson (1974) span the balance of the Maastrichtian, discussion of which is beyond the scope of this study.

Paleoecological Inferences

Previous Studies

The use of dinoflagellates for paleoecological interpretations is relatively new, with most studies having been

Figure 9. Stratigraphic ranges of the 23 dinoflagellate taxa common to Wilson (1974) and this study.

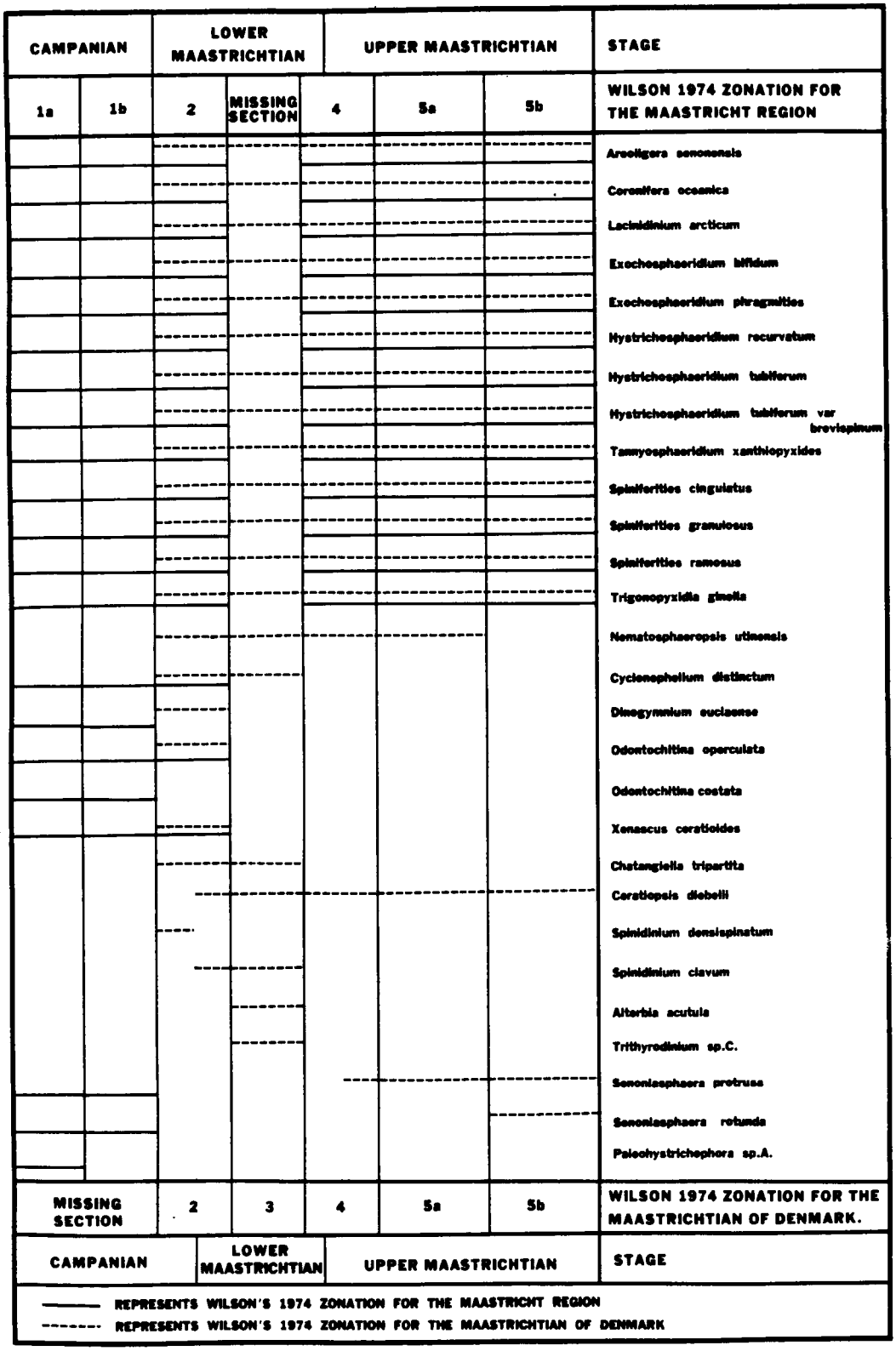

CAMPANIAN
LOWER MAASTRICHTIAN
UPPER MAASTRICHTIAN
STAGE
1a
1b
2
MISSING SECTION
4
5a
5b
WILSON 1974 ZONATION FOR THE MAASTRICHT REGION
Arcoligera senonensis
Coronifera oceanica
Lacinidinium arcticum
Exechosphaeridium bifidum
Exechosphaeridium phragmites
Hystrichosphaeridium recurvatum
Hystrichosphaeridium tubiferum
Hystrichosphaeridium tubiferum var brevispinum
Tanyosphaeridium xanthiopyxides
Spiniferites cingulatus
Spiniferites granulosus
Spiniferites ramosus
Trigonopyxidia ginella
Nematosphaeropsis utinensis
Cyclonephelium distinctum
Dinogymnium euclaense
Odontochitina operculata
Odontochitina costata
Xenascus ceratioides
Chatangiella tripartita
Carstiopsis diebelii
Spinidinium densispinatum
Spinidinium clavum
Alterbia acutula
Trithyrodinium sp.C.
Senoniasphaera protrusa
Senoniasphaera rotunda
Paleohystrichophora sp.A.
MISSING SECTION
2
3
4
5a
5b
WILSON 1974 ZONATION FOR THE MAASTRICHTIAN OF DENMARK.
CAMPANIAN
LOWER MAASTRICHTIAN
UPPER MAASTRICHTIAN
STAGE
REPRESENTS WILSON'S 1974 ZONATION FOR THE MAASTRICHT REGION
REPRESENTS WILSON'S 1974 ZONATION FOR THE MAASTRICHTIAN OF DENMARK

Figure 9

Table 3. The Northeast Texas samples examined (this thesis) have thirty taxa in common with the Wilson 1974 study.

WILSON 1974	CURRENT NAME AS USED IN THIS THESIS
Areoligera senonensis	Areoligera senonensis
Dinogymnium denticulatum	Dinogymnium denticulatum
D. euclaensis	D. euclaensis
Australiella balmei	Spinidinium clavum
A. acutula	Alterbia acutula
A. tripartita	Chatangiella tripartita
A. hvidensis	Spinidinium densispinatum
Diconodinium parvum	Lacinidinium arcticum
Deflandre diebelii	Ceratiopsis diebelii
Trithyrodinium suspectum	Trithyrodinium Sp. C.
Phobenocysta ceratioides	Xenascus ceratioides
Odontochitina operculata	Odontochitina operculata
O. costata	O. costata
Svalbardella parva	Paleocystadinium Sp. A.
Senoniasphaera protrusa	Senoniasphaera protrusa
S. rotunda	S. rotunda
Cyclonephellum distinctum	Cyclonephellum distinctum
Paleohystrichophora infuserioidies	Paleohystrichophora Sp. A.
Exochosphaeridium bifidum	Exochosphaeridium bifidum
E. phragmities	E. phragmities
Spiniferities ramosus	Spiniferities ramosus
Coronifera oceanica	Coronifera oceanica
Prolixosphaeridium xanthiopyxides	Tannyosphaeridium xanthiopyxides
Hystrichosphaeridium recurvatum	Hystrichosphaeridium recurvatum
Spiniferities cingulatus	Spiniferities cingulatus
Trigonopyxidia ginella	Trigonopyxidia ginella

Table 3 (Continued)

WILSON 1974	CURRENT NAME AS USED IN THIS THESIS
Spiniferities ramosus var. granosus	Spiniferities granulosus
Hystrichosphaeridium tubiferum var brevispinum	Hystrichosphaeridium tubiferum var brevispinium
Cannosphaeropsis utinensis	Nematosphaeropsis utinensis
Hystrichosphaeridium tubiferum	Hystrichosphaeridium tubiferum

conducted within the last twenty years. Wall et al (1977), in a study of modern marine sediments from the North and South Atlantic Ocean as well as the Gulf of Mexico, recognized that species richness increased with an increasing distance from shoreline. Goodman (1979) in a study conducted on Eocene strata of the Atlantic Coastal Plain, compared species dominance versus species diversity. Species dominance is defined as the sum of the two most abundant species within a given count, divided by the total observed specimens. Goodman defined species diversity as the total number of species encountered within the given count. In this thesis "taxonomic richness" is used as equivalent to "species diversity" in the sense of Goodman (1979). Goodman (1979) interpreted samples containing high species dominance and low taxonomic richness (species diversity) as being related to a more inshore environment; conversely, samples that

contain low species dominance and high taxonomic richness (species diversity) were interpreted as being from a more offshore environment.

Harland (1973) studies a Cretaceous section in Alberta Canada. A comparison was done on each sample contrasting the total number of specimens belonging to the family Gonyaulacaceae, versus the total number of specimens belonging to the family Peridiniaceae. This relationship was termed the Gonyaulacacean ratio, or (G/P)ratio. Samples with a relatively high (G/P)ratio were interpreted as coming from sediments indicating open marine conditions. Conversely, samples containing a relatively low (G/P)ratio were interpreted as coming from sediments indicating a more restricted, possibly brackish environment. Appendix E shows the grouping of dinoflagellate cysts by family used in this study.

Downie, et al (1971), in a study of Early Eocene strata in southeastern England, recognized reoccurring species associations and their affinity to particular lithologies. In that study, the authors were able to recognize three distinct species associations which were named after the dominant genus. The Spiniferites-Areoligera association was interpreted as being associated with sediments deposited in open marine environments. This association would probably compare with the high Gonyaulacacean ratio of Harland (1973) due to the dominance of Spiniferites and Areoligera, both genera in the Gonyaulacaceae. The

Wetzeliella association of Downie et al (1971) would probably compare with the low Gonyaulacacean ratio of Harland (1973), since the Wetzeliella association is dominated by species of Wetzeliella and Deflandrea, both genera of the Peridiniaceae. The Wetzeliella association was interpreted as originating in sediments deposited in a restricted, estuarine or brackish environment. In both cases, there is a parallel between the species association, their (G/P)ratio, and the inferred depositional environment. The third association of Downie, et al (1971), the Michrystridium association, was interpreted as representing deposition in an inner neritic or transitional environment.

Cluster Analysis

A cluster analysis was applied to the data in an attempt to group the samples objectively on the basis of dinoflagellate composition. For ease of interpretation, a dendrogram was used to organize the matricies of coefficients into easily visualized cluster groups (Sokal and Sneath, 1963).

Clapham (1972), comparing different statistical analyses available in palynological studies, clearly differentiates data formats and statistical tests on the basis of information sought by the investigator. Analytical mode is most critical in yielding biostratigraphically significant inferences on one hand, and in making paleoecological judgments on the other. The R-mode, with a

numerical count (taxa) matrix, shows relationships between taxa and assumes a normal distribution. The Q-mode, with a numerical count (taxa) matrix, shows relationships between samples, and there is no assumption of normality required. In Q-mode analysis, which does not necessarily have biological or geological significance, variation about the mean is due to non-random differences in rates of palynomorph production (Clapham 1972).

Clapham (1972) shows both common and rare palynomorph taxa, in this case pollen and spores, are normally distributed or nearly so. With an assumption of normally distributed taxa, an R-mode analysis can be applied to the data set.

Two methods of clustering (the Centroid method and Ward's method) are available in the Statistical Analysis System (SAS) computer facility at the University of Tennessee. Both methods are based on Euclidean distance as a means of measure, and both produce matrices of coefficients that can be visualized by the use of a dendrogram.

The SAS User's Guide (1982, p.423) describes the cluster procedure and Ward's method as follows:

> "CLUSTER uses three standard agglomerative hierarchial clustering algorithms. Each observation begins in a cluster by itself. The two closest clusters are merged to form a new cluster replacing the two old clusters. Merging of the two closest clusters is repeated until only one cluster is left. The algorithms used by CLUSTER differ in how the distance between two clusters is computed."

> "In Ward's method the distance between two clusters is the sum of squares between the two clusters added up over all the variables. At each generation, the within-cluster sum of squares is minimized over all partitions (an exhaustive set of disjoint clusters) obtainable by merging two clusters from the previous generation. Ward's method tends to join clusters with a small number of observations, and is biased toward producing clusters with roughly the same number of observations."

Ward's clustering method was used to cluster the data using the original counts of species composition. The first dendrogram (Fig. 10) shows the grouping of samples based on absolute number of specimens for each dinoflagellate taxon.

As noted above, Ward's clustering method differentially weights samples with small numbers of observations. Samples 15, 88, and 4, with 54, 7 and 42 specimens respectively, are clustered together; these samples have counts of total specimens substantially lower than the minimum count of 300 considered statistically reliable. Samples 15, 88, and 4, were removed from the data based and the cluster analysis was run again.

In the second run of the cluster analysis, all samples used in the run have comparable values for total specimen count (generally 300 or more); their species abundances dictate the observed clustering of the samples. The second dendrogram could be easily divided into 5 groups (Fig. 11). The Euclidean distance measure used in the clustering of the samples weighs the groupings on the basis of one or two species with high abundances in the samples, thus other species relationships are masked within a sample. For

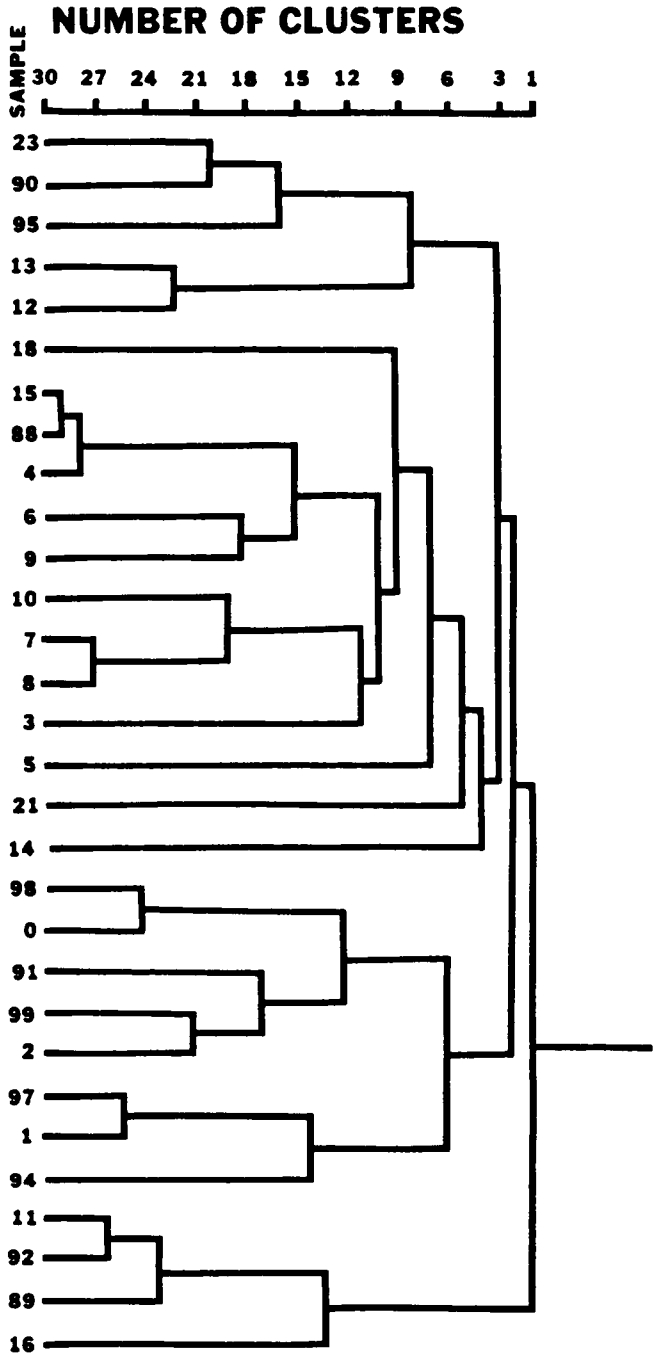


Figure 10. Cluster-analysis dendrogram using the original counts. Note the clustering of samples 4, 15 and 88.

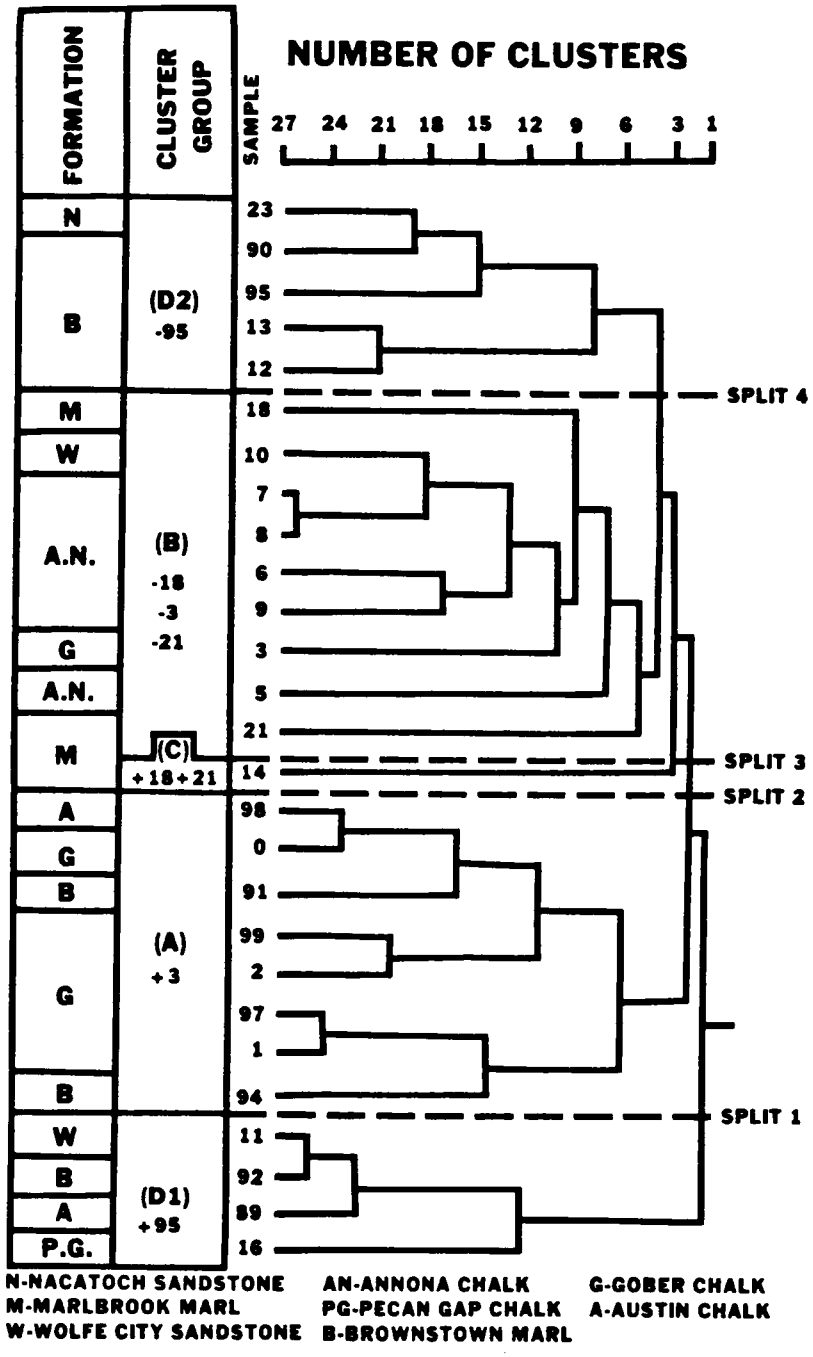


Figure 11. Cluster-analysis dendrogram. Note that samples 4, 15 and 88 were excluded because of their low (less than 300 specimen) counts.

example if for some reason (preservation bias or processing technique) a particular species is overrepresented compared to a normal representation (with ideal preservation), that sample may cluster into a group based on an abnormal abundance of one or two species. Sokal and Rohlf (1969) suggests that data which are not normally distributed can be transformed to fit a normal distribution. A transformation commonly used when data are numerical counts, is the square-root transformation (Sokal 1969). A square-root transformation of the data prior to the clustering process should reduce the apparent bias toward clustering samples based on abnormally high abundances of one or two species.

A third cluster analysis was run using the square-root transformed data (Fig. 12). Comparison of the resulting dendrogram with run two shows that group A picked up sample 3, and group D_1 picked up sample 95. Another recognizable change is that cluster group C and B are now clearly two separate groupings, with cluster group C containing all three of the Marlbrook Marl samples. Each of the cluster groups will be looked at in detail in the following section. An additional dendogram more accurately illustrated the position of the individual nodes in relation to their calculated coefficient of dissimilarity between clusters (Fig. 13).

Cluster Interpretation

Samples grouped by the cluster analyst using the set of square-root transformed data, provide a basis for

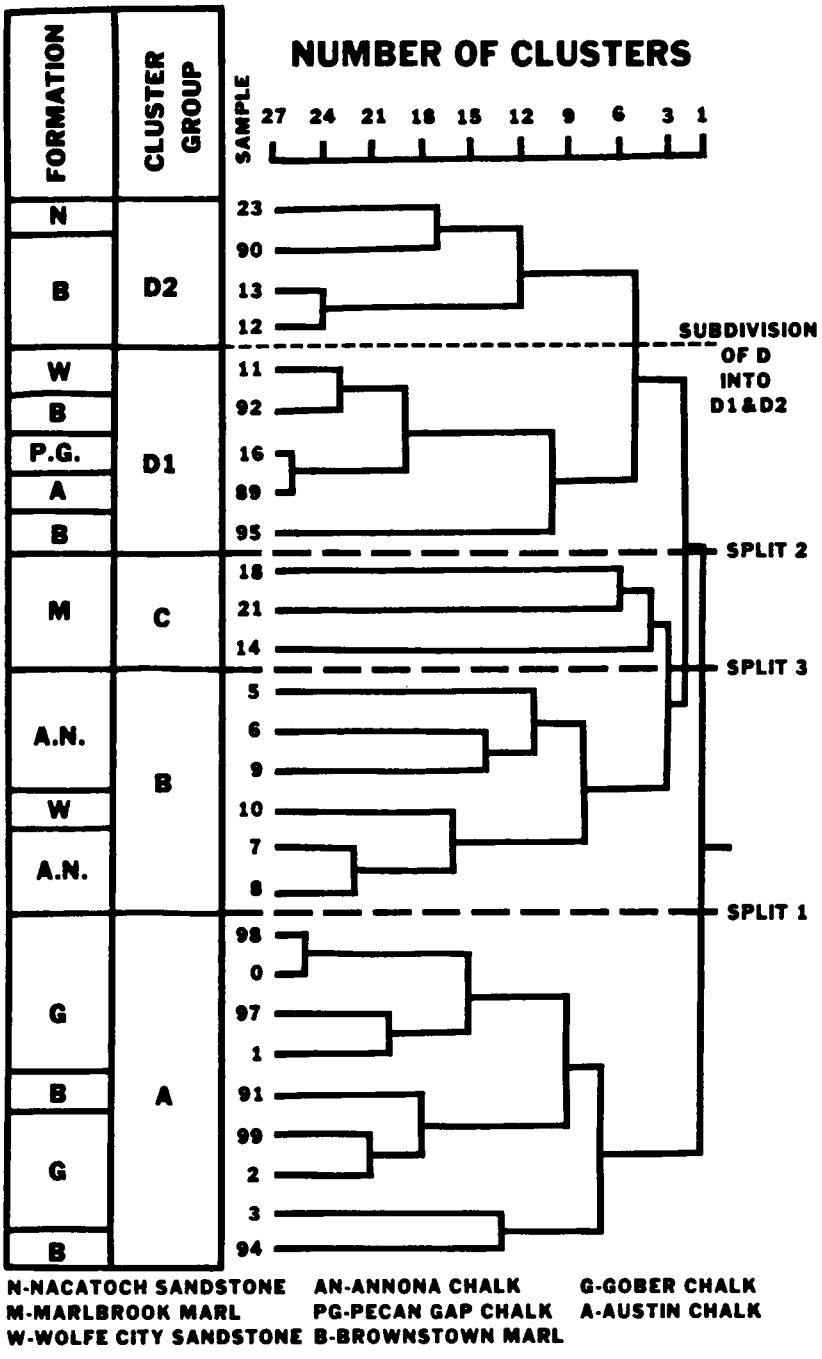


Figure 12. Cluster analysis dendrogram based upon a square-root transformation of the data set plotted with the number of clusters within an aggregation for scale. Note the four distinct divisions (A, B, C, and D), and one subdivision (D_1 & D_2).

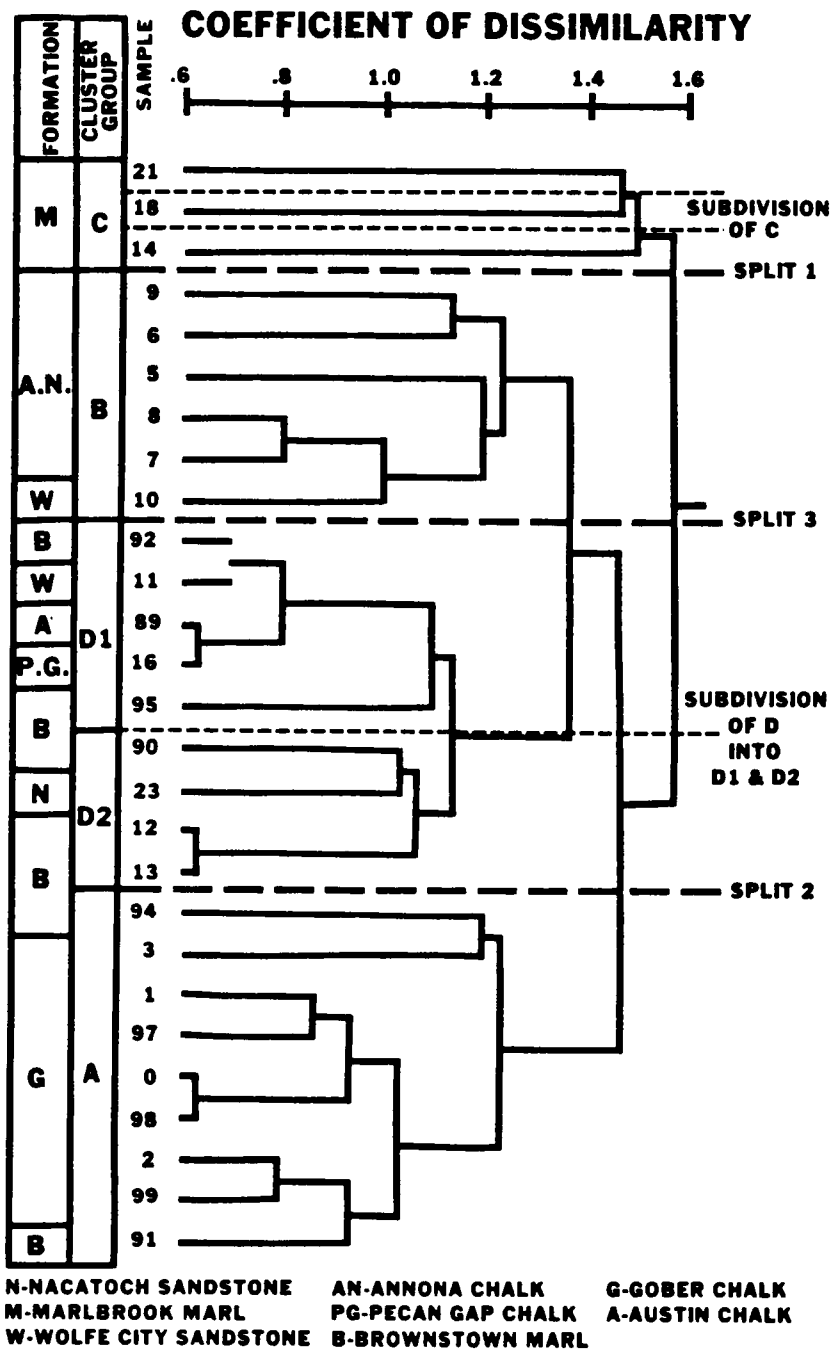


Figure 13. Cluster-analysis dendrogram using the square-root transformation of the data set plotted with the coefficient of dissimilarity for scale.

paleoecologic interpretation. Inspection of the dendrogram (Fig. 11) shows four distince divisions and one subdivision. There is relatively good correlation among the samples within a cluster, and the formations from which the samples were collected. All seven samples representing the Gober Chalk cluster in group A, and all five Annona Chalk samples cluster in group B. Cluster group C contains all the Marlbrook Marl samples, while cluster group D is primarily composed of samples from the Brownstown Marl. When the cluster groups are analyzed using species dominance and taxonomic richness (Fig. 14), (G/P) ratio (Fig. 15), and family association (Fig. 16), there appeared to be some correlation between the values derived from the method of analysis and the samples within a cluster group. The sedimentary environments associated with a particular formation, correspond with key limiting environmental factors that controlled the distribution of the dinoflagellate assemblages.

The cluster groups will be characterized in terms of: (1) their association with distinctive and lithology and their related depositional environment; (2) species dominance and taxonomic richness (species diversity in the sense of Goodman 1979); (3) (G/P) ratio (Harland 1973); and (4) D.X.O. percent. (D.X.O. percent represents the percent of the species not belonging to the family Gonyaulacaceae or Peridiniaceae, some of the more common taxa include species

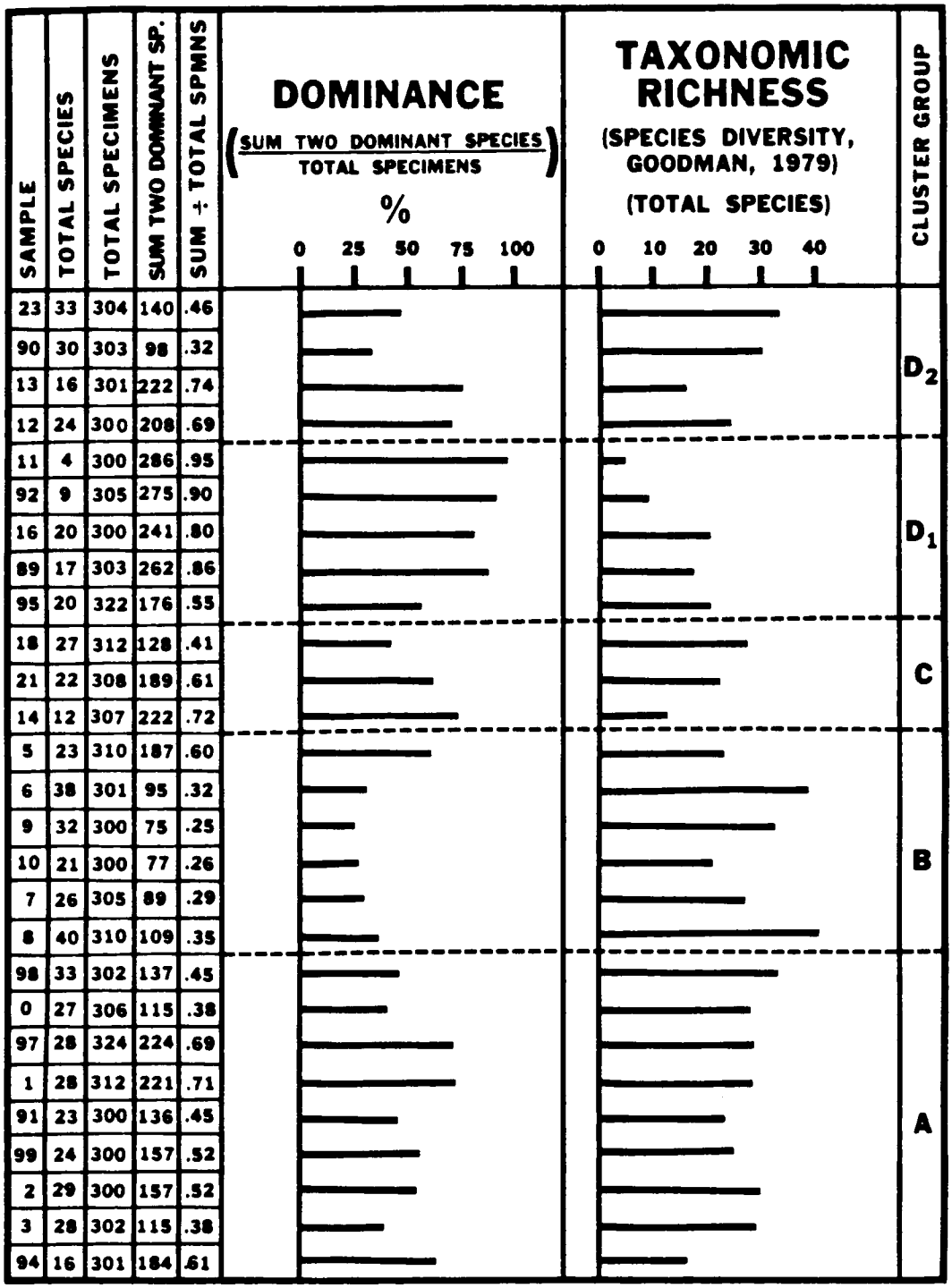
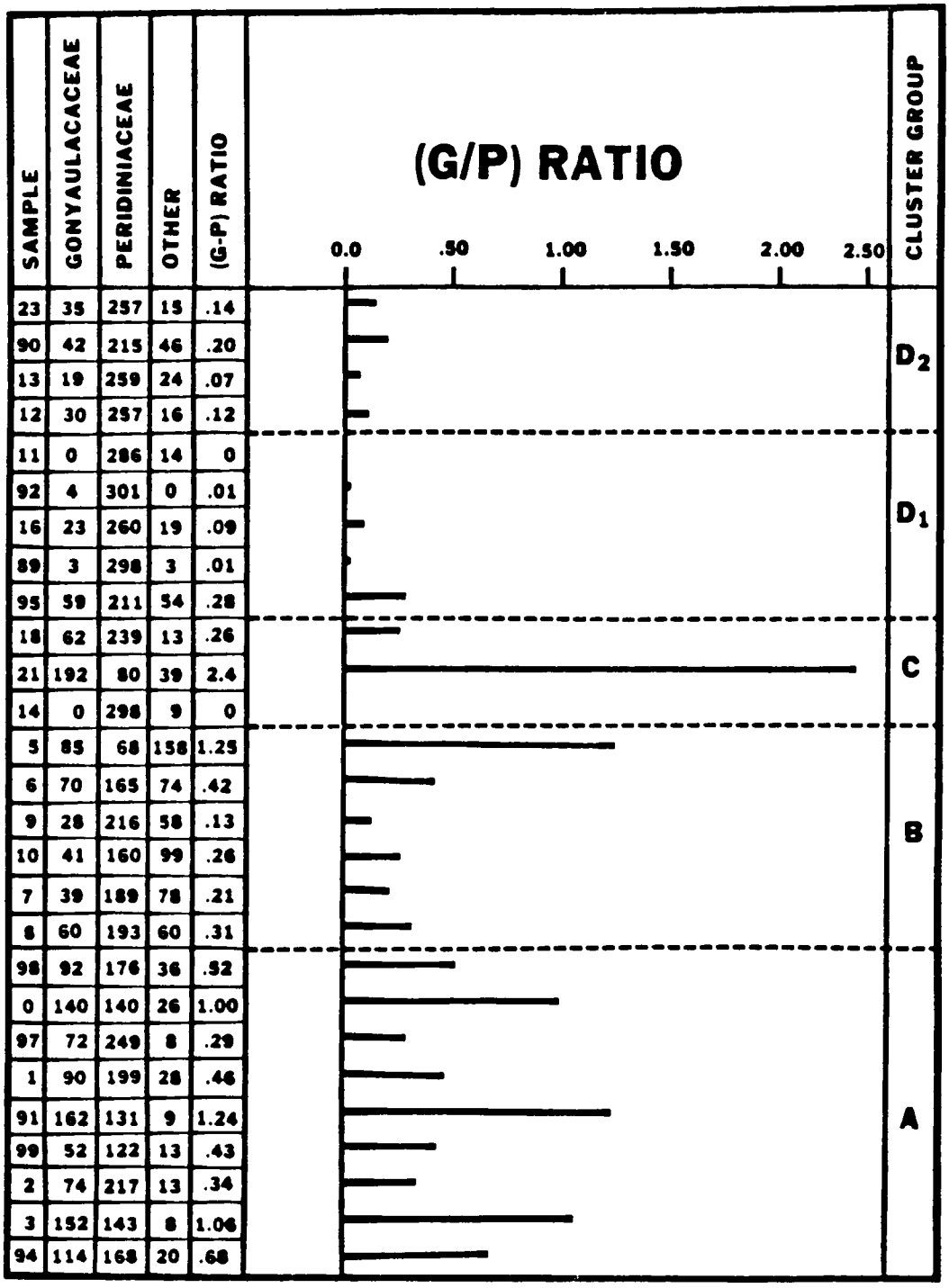


Figure 14. Species dominance and taxonomic richness. Samples are arranged according to clusters produced by using square-root transformation of the data set.



SAMPLE	GONYAULACACEAE	PERIDINIACEAE	OTHER	(G-P) RATIO	CLUSTER GROUP
23	35	257	15	.14	D_2
90	42	215	46	.20	
13	19	259	24	.07	
12	30	257	16	.12	
11	0	286	14	0	D_1
92	4	301	0	.01	
16	23	260	19	.09	
89	3	298	3	.01	
95	59	211	54	.28	
18	62	239	13	.26	C
21	192	80	39	2.4	
14	0	298	9	0	
5	85	68	158	1.25	B
6	70	165	74	.42	
9	28	216	58	.13	
10	41	160	99	.26	
7	39	189	78	.21	
8	60	193	60	.31	
98	92	176	36	.52	A
0	140	140	26	1.00	
97	72	249	8	.29	
1	90	199	28	.46	
91	162	131	9	1.24	
99	52	122	13	.43	
2	74	217	13	.34	
3	152	143	8	1.06	
94	114	168	20	.68	

Figure 15. (G/P) ratio of cysts assigned to the Gonyaulacaceae (G) to the cysts of the Peridiniaceae (P). The samples are arranged according to the clusters produced using the square-root transformed data set.

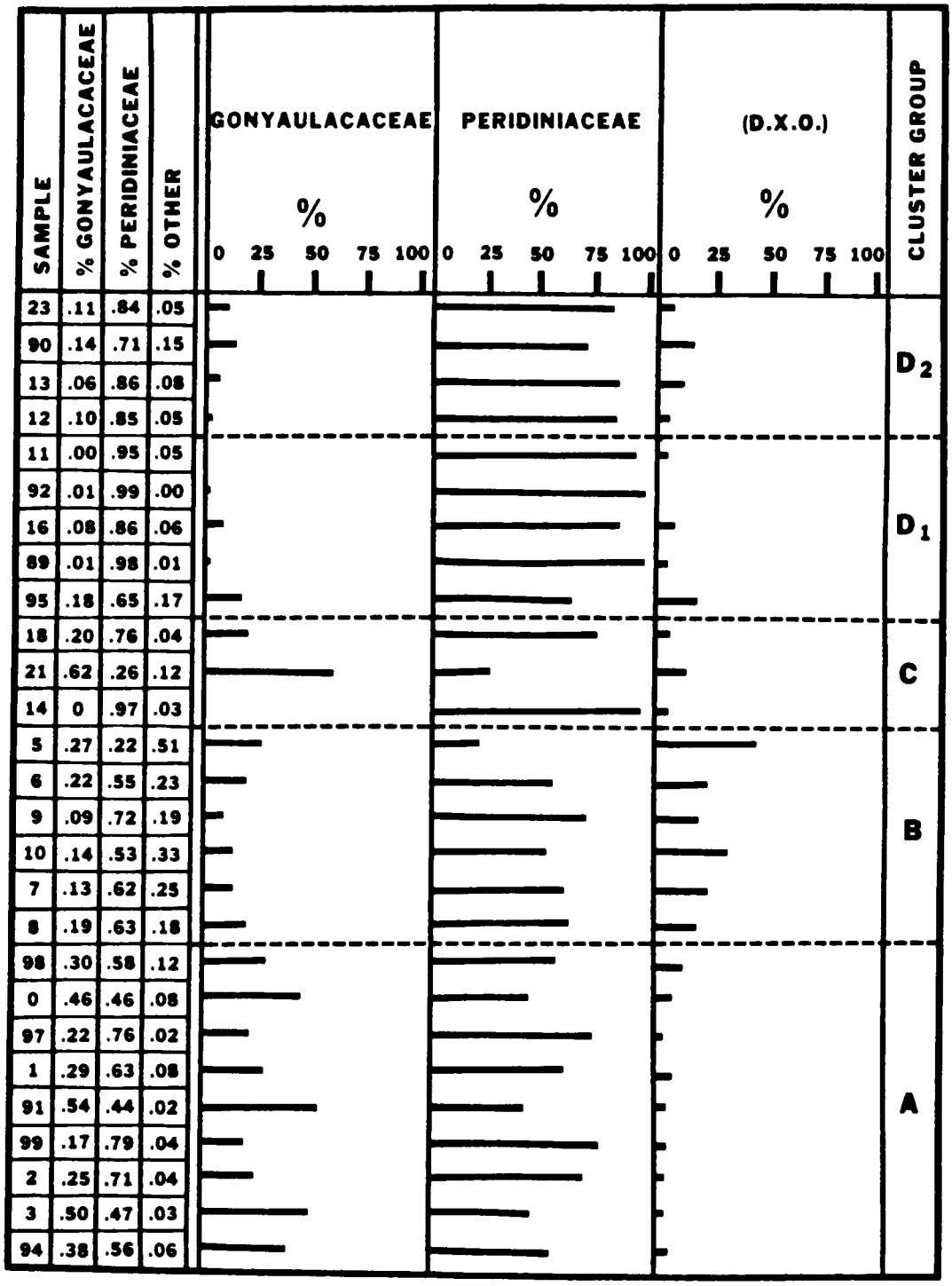


Figure 16. Graphic display of comparative representation in percent of species belonging to the Peridiniaceae, Gonyaulacaceae and (DXO) group. (DXO) group includes palynomorphs assigned to the genera Dinogymnium, Xenascus, and Odontochitina.

belonging to the genera Dinogymnium, Xenascus and Odontochitina.)

Cluster Group A Cluster group A contains all seven

Cluster Group A Cluster group A contains all seven

samples of the Gober Chalk plus two samples of the Brownstown Marl. This cluster group is characterized by moderate species dominance, high taxonomic richness, and high (G/P) ratio. The species responsible for the moderate species dominance observed in cluster group A are Palaeohystrichophora infusoroides and Spinidinium echinoideum of the Peridiniaceae along with Spiniferities ramosus and S. cingulatus of the Gonyaulacaceae.

The deposition of chalk delineates the limit of terrigeneous influence. Along with a lithological change from marl to chalk, there is a noticeable increase in the (G/P) ratio. It is apparent that members of the Gonyaulacaceae had a distinct preference for the clear water deposition overlying the Gober Chalk. Harland (1973) suggests that high (G/P) ratios indicate open marine conditions. Selznic (1983), based on a study of foraminifera, has suggested that the Brownstown Marl and the Gober Chalk were both deposited in water of the same depth middle neritic (50-100 meters), and there was no real change in bathymetry from one depositional environment to the other. Therefore, one controlling factor in limiting the existence of species belonging to the Gonyaulacaceae appears to be turbidity.

Species distribution characteristics of cluster group A are:

1. A dominance in *Palaeohystrichophora infusorioides*.
2. Species common to cluster group A samples.

   Exochosphaeridium bifidum
   Spiniferites ramosus
   S. cingulatus
   Hystrichosphaeridium palmatum
   Odontochitina porifera
   Spinidinium sp 1.
   Rottnestia borrussica
   Florentinia clavigera

3. Species restricted to group A samples

   Apteodinium cribrosum
   Senoniasphaera rotunda
   * *S. protrusa*
   Altberbia acutula
   Unknown C
   Florentinia sp. 1
   * *Florentinia laciniata*
   * *Operculodinium* sp.
   * *Surculosphaeridium* sp.
   Diphyes sp.
   Unknown D
   Canningia sp.1
   Nematosphaeropsis pusulosa
   Eatonicysta sp.

* Indicates a limited occurrence outside cluster group.

Cluster Group B Cluster group B contains palynomorphs from the five samples collected from the Annona Chalk, and one Wolfe City Sandstone sample. This cluster group is

characterized by low species dominance, high taxonomic richness moderate (G/P) ratio and consistently high D.X.O. percent.

The deposition of the Annona Chalk seaward of the sand beds formed along the flanks of topographic highs during the Campanian, has been proposed by Thompson (1983). Thompson interpreted the asymmetric distribution of the sand and chalk units with both thinning to the east, to reflect a clockwise current pattern within the shelf area. Earlier, Stehli and Creath (1972), proposed a system of marine current to account for the re-working of clastic material across the shelf and into the topographic lows, to form marl deposits. I consider the lithology of the Annona Chalk to indicate clear water deposition and delineate the limit of clastic influence from the west.

On the basis of foraminifera, Selznic (1983) proposed an inner to middle neritic depositional environment for the Annona Chalk. He recognized two phases of deposition, a lower regressive phase and an upper transgressive phase. In line with the proposed position of the Annona Chalk along the flanks of topographic highs, it is the opinion of Thompson (personal communication) that parts of the Annona Chalk represent the shallowest chalk deposition within the study area.

The implication of clear water deposition for chalk is supported by the low species dominance coupled with high

taxonomic richness recognized within cluster group B. The value of the (G/P) ratio is relatively low, which according to Harland (1973) indicates a more restricted environment.

The mud-influenced Gober Chalk, associated with cluster group A, was characterized previously as having high species dominance. The periodic influx of terrigenous mud, however, did not suppress chalk deposition. Cluster group A has also been interpreted as being open marine based on a high (G/P) ratio. On the other hand, the Annona Chalk, cluster group B, is lithologically a massive limestone showing limited terrigenous muds and is characterized by low species dominance and high taxonomic richness. The low (G/P) ratio may support the contention of a restricted environment, apparently related to the deposition of the Annona Chalk in close proximity to the shoreline or under shallow marine conditions.

It is apparent upon examination of the specific taxa within cluster group B, that the environment was not favorable for taxa belonging to the Gonyaulacaceae yet was favorable to high taxonomic richness and low species dominance. This environment also favored the presence of species belonging to the Gymnodiniaceae, as indicated by a high (D.X.O.) percent (Fig. 15). Species of Dinogymnium, and to a lesser extent Xenascus and Odontochitina, comprise the (D.X.O.) group in all samples when present.

Thus, the special conditions under which the Annona Chalk was deposited and the corresponding dinoflagellate palynomorphs assembled were those indicating a near shore, shallow but clear water, inner to middle neritic marine environment with minimal clastic influence.

Species distribution characteristics of cluster group B are:

1. No single dominant species, but a general abundance of species belonging to the Peridiniaceae.
2. Species common to all the samples in cluster group B.

   Palaeohystrichophora infusorioidies

   Palaeohystrichophora sp.B

   Chatangiella granulifera

   C. victoriensis

   Dinogymnium acuminatum

   Spiniferites ramosus

   S. cingulatus

   Odontochitina operculata

   Xenascus ceratioides
3. Species restricted to cluster group B.

   Dinogymnium sp.1

   D. longicornis

   Aoria sp.

   D. digitum

   D. cerviculum

   D. sp.2

Deflandrea sp.3

Dinogymnium denticulatum

Oligosphaeridum sp.

Cluster group C. Cluster group C contains palynomorphs from all the samples representing the Marlbrook Marl. Species dominance is moderate to high; taxonomic richness is moderate to low; and the (G/P) ratio was the lowest encountered in this study with a value of zero, while sample (18) has the highest with a value of (2.4).

Inspection of the dendrogram (Fig. 13) shows that samples 18, 21, and 14 are joined together very late in the clustering sequence, indicating a low degree of similarity. A side by side comparison of the sample counts reveals that the samples are not grouped on the basis of species dominance (as for cluster sub-group D_1), or by species association (as for cluster groups A, B, and D_2). Cluster group C has only three species common to all the samples, Trithyrodinium sp.1., Laciniadinium arcticum and Odontochitina operculata, none of which are restricted to the cluster group or occur in any unusual abundance. Cluster group C appears to lack species commonly found in a high percentage in all the remaining samples. Palaeohystrichophora infusorioides, and Chatangiella victoriensis occur in virtually every sample other than those in cluster group C and often these species

are a factor in determining cluster groupings, so their presence (or absence) is significant. Another noticeable difference is that cluster group C has a different species assemblage representing the Peridiniaceae in the (G/P) ratio. Cluster groups A, B, D_1, and D_2 are characterized by species belonging to Palaeohystrichophora, Chatangiella and Spinidinium echinoideum. In contrast, cluster group C contains specimens of Alterbia sp.1, Alterbia sp.3, Trithyrodinium sp.1, unknown peridinioid 518, Spinidinium sp.B, S. densispinatum and Palaeocystodinium sp.1.

Since the species that have been discussed as missing in the Marlbrook Marl occur stratigraphically above the Marlbrook in the Nactoch Sandstone, one must look at factors in the environment to explain the species distribution. The lower contact of the Marlbrook is lithologically and paleontologically gradational with the underlying units according to Selznic (1983). In conjunction with foraminiferal evidence, and the presence of radiolarians, Selznic (1983) suggests an outer neritic depositional environment for the Marlbrook Marl. He considered the transgressive pulse during Marlbrook Marl deposition to be of a greater magnitude than the preceding transgressive regressive sequence.

Sample 14 of cluster group C, is characterized by high species dominance, low taxonomic richness and a (G/P) ratio value of zero, the lowest of all the samples within the study area. Sample 14 was collected very close to the Marlbrook

Marl - Pecan Gap Chalk contact (Thompson 1983 p. 189 showing sample locations), perhaps indicating a transitional environment similar to those described in cluster group D_1. Similarly the samples within cluster group D_1 were characterized by high dominance, low taxonomic richness and low (G/P) ratio. In cluster group D_1, the abundant Peridiniaceae species responsible for both the low (G/P) ratio and the high dominance value is Palaeohystrichophora sp.B, while in sample (14) the dominant species is Alterbia sp.3 which accounted for 56% of the specimens counted. The pattern is the same, but the species are different. The depositional environment surrounding the transition from the Pecan Gap Chalk into the Marlbrook marl has been described as outer neritic according to Selznic (1983), based on foraminifera. Selznic was able to recognize a rapid transgression from the Wolfe City sandstone, through the Pecan Gap chalk and into the Marlbrook Marl. The presence of Alterbia sp.3 as the dominant peridinioid in place of Palaeohystrichophora sp.B could possibly be related to water depth, while the low (G/P) Ratio indicates environmental instability or restriction.

Away from the transition zone between depositional environments, the Marlbrook Marl appears to follow a more recognizable pattern. I interpret the moderate dominance and relatively moderate taxonomic richness along with a high (G/P) ratio as evidence for a more open marine environment.

The extremely high (G/P) ratio may reflect open marine conditions with possibly an increased distance from shoreline, with continued sea transgression.

Species distribution characteristics of cluster group C are:

1. Species belonging to the Peridiniaceae accounting for the low (G/P) percent.

   Alterbia sp.3

   Spinidinium densispinatum

   Spinidinium sp.3

   Palaeocystodinium sp.1

   Alterbia sp.3

   Unknown peridinioid 518

   Trithyrodinium sp.1

2. Species common to all cluster group C samples.

   Trithyrodinium sp.1

   Laciniadinium arcticum

   Odontochitina operculata

3. Species not represented in cluster group C common to all other cluster groups.

   Palaeohystrichophora infusorioides

   Chatangiella victoriensis

Cluster group D. Cluster group D, is interpreted as representing the most restricted environment as far as being unfavorable to a rich taxonomic assemblage, and the presence of species belonging to the family Gonyaulacaceae. Within

cluster group D there are two distinct sub-groups, D_1 and D_2. Samples contained within cluster sub-group D_1 contain a species assemblage I am interpreting as indicating the more restricted of the two environments which may possibly be a more restricted facies equivalent of cluster sub-group D_2.

Cluster sub-group D_1 contains palynomorphs from two Brownstown Marl samples, and from one sample each of the Wolfe City Sandstone, Austin Chalk and Pecan Gap Chalk. Although there appears to be no correlation with the lithologic character of the samples, there is a good correlation within the cluster group using other methods of comparison. All samples show very high species dominance, low taxonomic richness and very low (G/P) ratio values.

There is one species that appears to dominate every sample within this cluster sub-group and is probably the reason for their being grouped together. This species is Palaeohystrichophora sp.B, which accounts for a minimum of 38% of the specimens in sample 95, to as much as 87% of the specimens in sample 11. The other cluster group with a high representation of Palaeohytrichophora sp.B, is cluster sub-group D_2. In group D_2, other species are also common such as Chatangiella granulifera, P. infusorioides, Spiniferites ramosus and Odontochitina operculata. Of these, only C. granulifera and P. infusorioides occur very rarely in cluster sub-group D_1.

Based on lithologic data, the Wolfe City Sandstone and the Brownstown Marl represent near shore, turbulent marine environments. Whereas in the case of the two chalk samples, the same line of reasoning may not lead to the same conclusion. Upon closer examination however, the two chalk samples were taken very close to the contact with an adjacent formation. Austin (sample 89), represents one of the calcareous mudstone interbeds of the Austin Chalk, very close to the gradational contact with the Brownstown Marl. The Pecan Gap (sample 16) was collected from the sandy Lavon Member near the contact with the Wolfe City Sandstone (Thompson personal communication, June 12, 1984). Palaeohystrichophora sp.B, appears to have been tolerant of a wide range of conditions as can be seen in the species distribution data and, under stressed conditions, dominates all other species. Palaeohystrichophora sp.B is present in all but five samples within this study area and has its greatest abundance in samples from stressed environments which are geographically restricted.

Cluster group B was interpreted as having a somewhat restricted environment, based on a low (G/P) ratio. Low counts on species belonging to the Gonyaulacaceae are paralleled by the presence of Palaeohystrichophora sp.B. Cluster group B consists of samples from the Annona Chalk which lithology indicates having a limited clastic influence. It is

postulated that the influx of terrigenous material is one source of stress affecting abundances of Palaeohystrichophora sp.B.

Cluster sub-group D_2 contains three samples from the Brownstown Marl and one sample from the Nacatoch Sandstone. This cluster sub-group has a similar dominance and taxonomic richness index to cluster group A, but the (G/P) ratio was significantly lower.

According to Selznic (1983), the Gober Chalk and parts of the Brownstown Marl were deposited during a marine transgression and subsequent regression. Foraminiferal assemblages in the Gober Chalk and the Brownstown Marl have relatively high planktonic to benthic ratios, with the marl yielding a higher average number of planktonic specimens than the chalks. Selznic postulated that an influx of terrigenous mud into the water column overlying the marl reduced the number of benthic species to those able to tolerate the resulting turbidity. Terrigenous mud is recognized in the deposition of the Brownstown Marl by the calcareous mudstone lithology. In response to the increased turbidity, the dinoflagellate assemblage shows moderate species dominance, moderate taxonomic richness and very low (G/P) ratio, which characterizes cluster sub-group D_2. Species responsible for the high dominance in cluster sub-group D_2 are Palaeohystrichophora sp. B and Chatangiella granulifera, both members of the Peridiniaceae. The species responsible for the moderate to high dominance observed in

cluster group A are Palaeohystrichophora infusorioides and Spinidinium echinoideum of the Peridiniaceae along with Spiniferites ramosus and S. cingulatus of the Gonyaulacaceae. At first it appeared that the dominance and diversity index was not differentiating between the two groups, but when the species responsible for the high dominance value are considered, it is clear that the two clusters are distinct.

Two species of Palaeohystrichophora are common throughout the study area, but achieve dominance in two different environments. Palaeohystrichophora infusorioides appears to achieve dominance in the chalks of cluster group A, suggesting a preference toward clear water deposition. Palaeohystrichophora sp. B achieves its highest dominance in cluster sub-group D_1 and second highest in cluster sub-group D_2.

Species distribution characteristics of cluster sub-group D_1 are:

1. Very high abundance of Palaeohystrichophora sp. B.
2. Very low representation of species belonging to the family Gonyaulacaceae.

Species distribution characteristics of cluster sub-group D_2 are:

1. General high abundance in: Chatangiella granulifera, Paleohystrichophora sp. B, and P. infusorioides.
2. Species common to all samples include: Odontochitina costata; Spiniferites ramosus, plus those mentioned above as highly abundant.

Dinoflagellate to Pollen and Spores as a Relative Measure of Distance from Shoreline

The ratio of dinoflagellates to pollen and spores has been used as a relative measure of proximity to shoreline. Several studies notably Davey and Rogers (1975) indicate that the ratio of dinoflagellates to pollen and spores will increase, with increasing distance from shoreline. In sample processing, residues were screened with a mesh size of 35 microns, and slides were prepared from the residue fraction greater than 35 microns. Because of the loss of the less than 35 micron fraction of the residues in the screening process (mostly pollen and spore grains), the dinoflatellate to pollen and spore ratio will be exaggerated. With this in mind, each sample was counted to a total of 100 palynomorphs which were divided into two separate groupings, dinoflagellates, or pollen and spores. The total number of dinoflagellate in the count of 100 palynomorphs are figured using three composite dip sections, Rockwall, Gober-Paris, and Clarksville (Fig. 17). The stratigraphic placement of the samples follows the sequence proposed by Thompson (1983), while their lateral position within the dip section is controlled by the dinoflagellate to pollen and spore ratio. The low ratios in each dip section are interpreted as indicating near shore environments. Whereas high ratios are interpreted as indicating a more offshore marine environment.

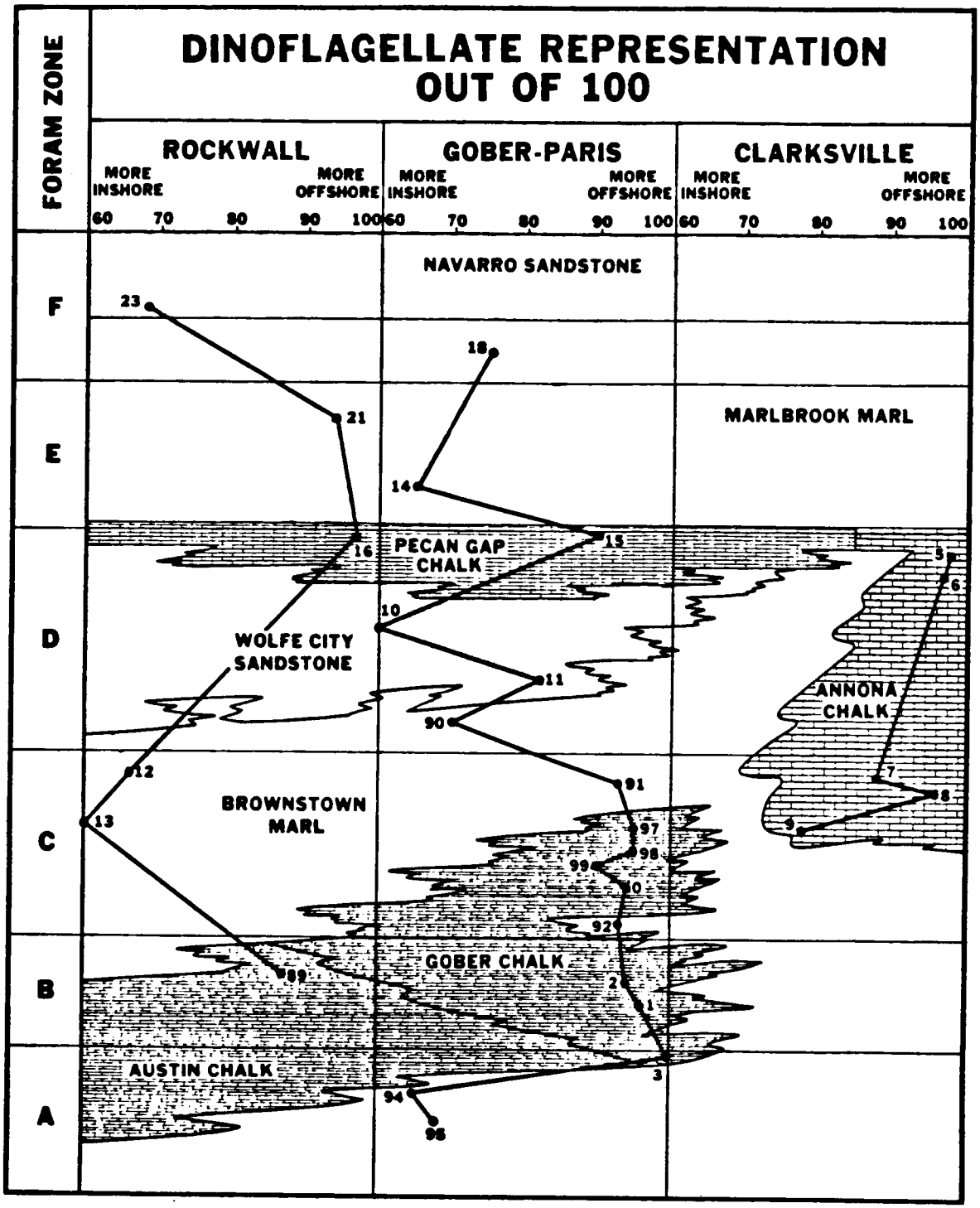


Figure 17. Dinoflagellate count out of 100 palynomorphs counted arrayed in three dip-sections, Rockwall, Gober-Paris, and Clarksville.

The Gober-Paris dip section has the most complete sample distribution vertically through the section. Four nearshore cycles separated by three offshore cycles can clearly be recognized in the Gober-Paris dip section. As in the species dominance and taxonomic richness index there appears to be a correlation between lithological character and the dinoflagellate to pollen and spore ratio. The high ratio values are associated with the chalk units and near by marl units, whereas the low ratio values are associated with the marls and sandstones (Fig. 18). The Brownstone Marl surrounding the Gober Chalk has relatively low dinoflagellate to pollen and spore ratios (samples 94, 95, 90, 12 and 13). The samples representing the Gober Chalk (01, 02, 00, 99, 98, & 97) have relatively high ratios indicating a more offshore environment. The more inshore environment indicated by the Brownstown Marl overlying the Gober Chalk continues into the Wolfe City sandstone (samples 10 and 11), with low ratio values, followed by an increasing distance from shoreline trend into the overlying Pecan Gap Chalk (samples 15 and 16). The final cycle begins with a more inshore trend in the early Marlbrook Marl (sample 14), increasing in distance from shoreline within the Marlbrook (sample 21), and then a final inshore trend (sample 18) terminating in the Nacatoch sandstone (sample 23).

The Clarksville dip section contains only Annona Chalk samples. The trend within the Annona Chalk goes from

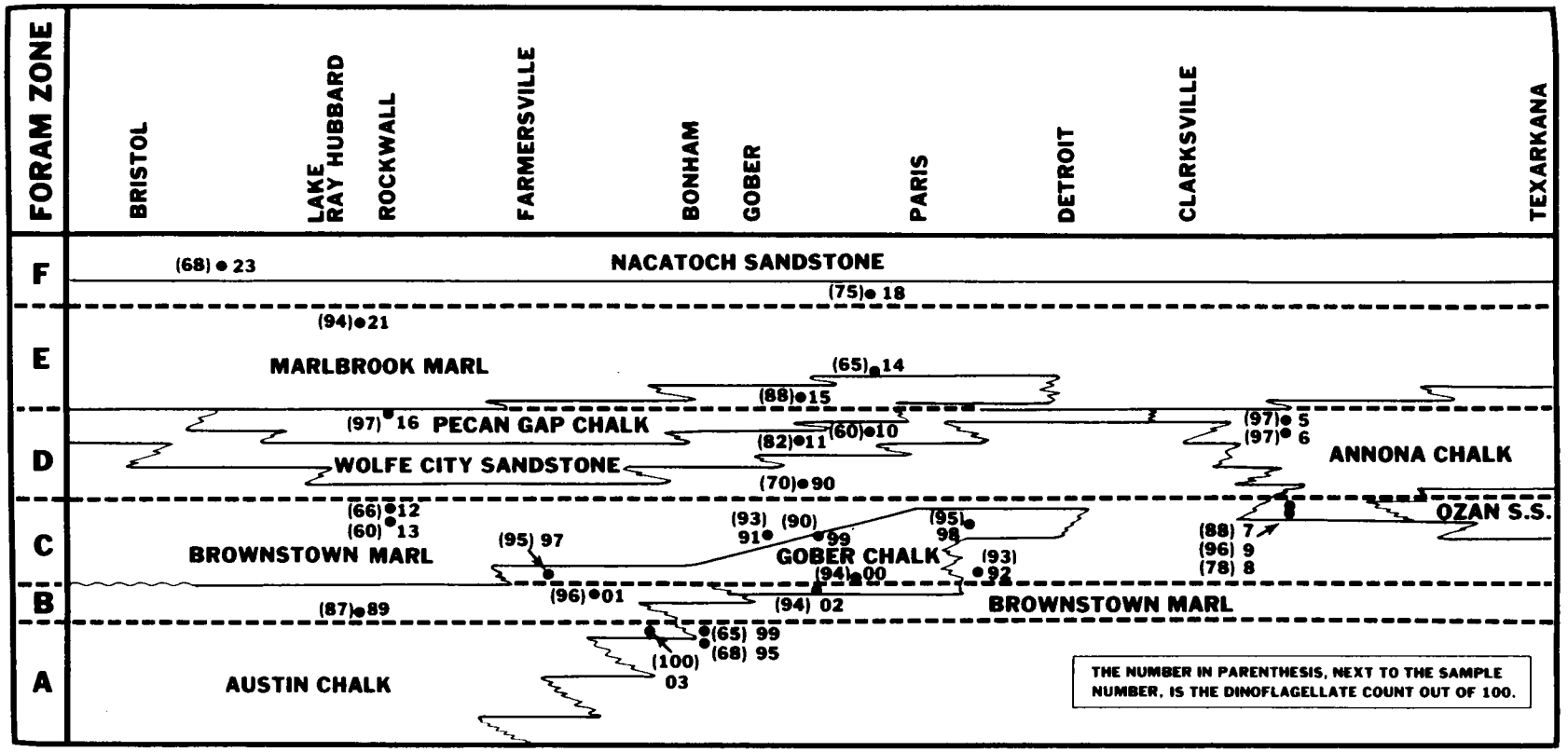


Figure 18. Lithofacies distribution map showing sample locations and dinoflagellate representation out of 100 palynomorphs counted per sample.

a more inshore to a more offshore environment up through the section with the furthest offshore section being correlative with the Pecan Gap Chalk.

The inshore - offshore interpretations given above appear to coincide with the other interpretations of this thesis, except for the placement of Gober Chalk further offshore than the Brownstown Marl. One explanation might be that the pollen and spore distribution within the Brownstown-Gober system is constant, but the clear water associated with the deposition of the Gober Chalk may have supported an increase in dinoflagellate production, thus increasing the dinoflagellate percent. This line of reasoning holds for the Annona Chalk, previously interpreted as a near shore environment based on the (G/P) ratio. Again, there may have been a normal pollen and spore distribution but increased dinoflagellate production due to clear water deposition accounting for high dinoflagellate to pollen and spore values.

CHAPTER 4

SUMMARY AND CONCLUSIONS

Biostratigraphy

Of two previous palynological studies available for comparison, only the Zaitzeff and Cross (1966) study dealt with geographically related rocks. Wilson (1974) examined the type Campanian and Maestrichtian sections in Holland, Belgium and Denmark.

A possible lithostratigraphic problem may have been introduced in the Zaitzeff and Cross (1966) study because problematical strata may have been included which are no longer regarded as occurring within the Campanian and Maestrichtian boundaries. Twenty-three of the fifty-six dinoflagellate taxa Zaitseff and Cross used to zone the Maestrichtian are shared in common with the list of taxa identified in the present study. Both lithological and paleontological evidence supports the conclusion that the Neylandville Marl portion of zone A belongs to the Taylor Group and is Campanian in age.

Comparison with the Wilson (1974) study is limited to zones one and two of Wilson's five dinoflagellate zones. The thirty taxa found in common with this study are figured in Figure 9. Wilson uses the first stratigraphic occurrence of *Deflandre diebelli*, and *Austraiella balmei* and the last

stratigraphic occurrence of Austraiella hvidensis, (Ceratiopsis diebelii, Spinidinium clavum and Spinidinium densispinatum this study) to mark the Campanian-Maestrichtaan boundary in the Maestrichtian of Denmark. In the Northeast Texas section both Ceratiopsis dibelii, and Spinidinium clavum extend into the Campanian, while spinidinium densispinatum continues into the Maastrichtian. Wilson uses the last stratigraphic occurrence of Odontochitina operculata, O. costata, Senoniasphaera protrusa, and S. rotunda to mark the Campanian-Maestrichtian boundary in the Maestricht region. In Northeast Texas, both Odontochitina costata and O. operculata extend into the Maestrichtian, while Senoniasphaera protrusa and S. rotunda end within the Campanian.

Paleoecology

Cluster analysis has shown five distinct dinoflagellate assemblages considered to be environmentally controlled. Four of these are coincident with a particular lithology, i.e., the Annona Gober, Brownstown and Marlbrook formations. The fifth is considered to be a transitional assemblage.

The cluster group B of dinoflagellates is primarily associated with the Annona depositional environment, a shallow, inner to middle neritic, clear water environment with little clastic influence. The environment of deposition of the Annona chalk was characterized by low species dominance, high taxonomic richness, moderate to low (G/P) ratio and a

high (D.X.O.) percentage. Characteristic species of the Annona environment includes a variety of Dinogymnium species, Xenascus ceratioides, Odontochitina operculata Chatangiella victoriensis, C. granulifera, Palaeohystrichophora sp.B, P. infusorioides and Spiniferites ramosus. The moderate (G/P) ratio reflects a somewhat restricted environment associated with a close proximity to shoreline.

The dinoflagellate grouping (A) within the Gober depositional environment is inferred to have lived in middle neritic, clear water in which deposition of chalk was interrupted by periodic pulses of mud which are represented by calcareous mudstone interbeds. The sedimentary environment of the Gober Chalk supported moderate to high species dominance, moderate to high taxonomic richness, high (G/P) ratio, and a low (D.X.O.) percent. Characteristic species of the Gober chalk include abundant Palaeohystrichophora infusorioides, and common Exochosphaeridium, bifidum Spinidinium sp. 1, Spiniferites ramosus, S. cingulatus and Hystrichosphaeridium sp. 1. An open marine environment of the Gober chalk is indicated by a high (G/P) percent. Periodic influx of mud, however, maintains high species dominance.

Dinoflagellates in the cluster sub-group D_2 lived within the Brownstown depositional environment, one of middle neritic, calcareous mudstone deposition with a high degree of terrigenous influence. The environment in which the deposition of the Brownstown Marl took place supported high species

dominance, moderate to low taxonomic richness, very low (G/P) ratio and a low (D.X.O.) percent. Species characterizing the Brownstown type environment are dominant Palaeohystrichophora sp. B, and common Chatangiella granulifera, P. infusorioides and Odontochitina costata. Marine conditions during the deposition of the Brownstown Marl are somewhat restricted. This may be due, in part, to an increase in clastic influence.

Cluster group C includes dinoflagellates that lived in the Marlbrook Marl environment,lithologically similar to that of the Brownstown Marl, both being associated with the deposition of a calcareous mudstone. From paleontologic evidence, Selznic (1983) suggested that the Marlbrook Marl was deposited in much deeper, outer neritic (100-200 M) water, which supported a different floral assemblage than that of the Brownstown. Unlike the other clastic influenced environments, the Marlbrook has no Palaeohystrichophora infusorioides and specimens of Palaeohystrichophora sp. B are rare. Other peridiniacean taxa usually found associated with muddy or stressed environments are missing. Instead, Alterbia sp. 3, Alterbia sp. 1, Trithyrodinium sp. 1, Palaeocystadinium sp. 1, and Spinidinium sp. 2 make up a significant proportion of the species of Peridiniaceae present. It is possible that these taxa are indicative of deeper water where terrigenous influence was extended. The high (G/P)

ratio indicates that bottom currents were responsible for the increased turbidity. Thus, the unaffected surface waters could still support numbers of open marine indicator species as in sample 21.

Cluster sub-group D_1 of dinoflagellates are characterized by an extremely high dominance of Palaeohystrichophora sp B. Of the five samples assigned to this environment type, three samples (11, 16, and 89) were taken close to a lithologic transition between two depositional environments, thus indicating an unstable environment between two depositional environments.

In Figure 19, the five depositional environments discussed above are arrayed in a postulated paleoenvironmental framework to show inter-relationships of marine conditions, including water depth, the environments of deposition, and the position of lithologic samples from which the environmental inferences were derived for the dinoflagellate assemblages.

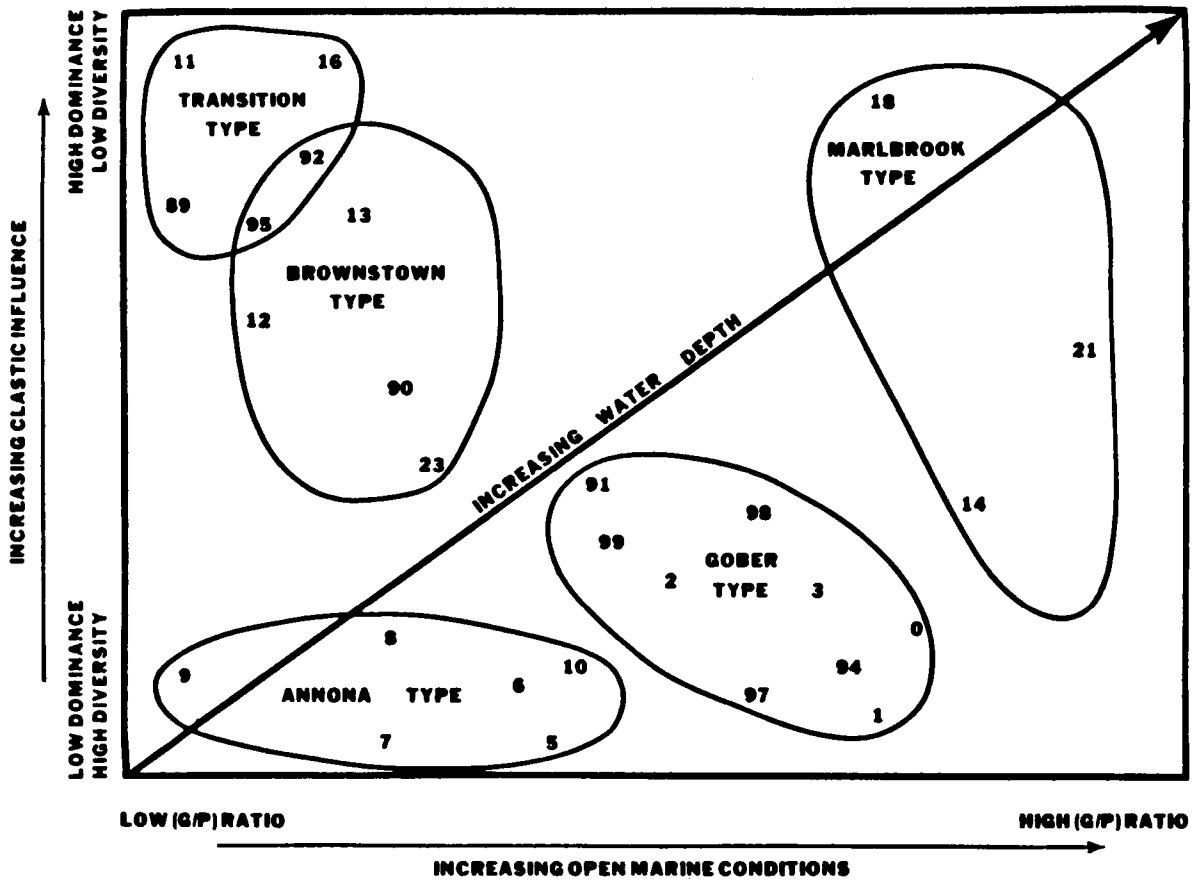


Figure 19. Distribution of samples in relation to the postulated type environments.

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APPENDICES

Appendix A

Sample Locations

The sample numbers used in this thesis are in the parenthesis following the corresponding sample number Thompson (1983), used in his study.

7-24-7-4d (23) Nacatoch Sandstone. In low outcrop by the side of an unpaved road 180 m east of farm road 1390, 4.8 km northwest of Scurry, Kaufman County, Scurry quadrangle.

9-17-9-3 (18) Marlbrook Marl. In south side of road cut on Texas highway 24 just north of Cooper (1.2 km north of town center) at old Southern Pacific Arilroad intersection, Delta County, Cooper North quadrangle.

11-19-7-4a (21) Marlbrook Marl. In deep stream cut just south of farm road 2755, 1.9 km west of farm road 1138. About 5.63 km southeast of Lavon, Collin County, Lavon quadrangle.

8-10-9-7e (14) Marlbrook Marl. Very close to the Pecan Gap Chalk (see diagram in Thompson 83 p. 189 showing locality 8-10-9-7 and sample locations). Locality in eroding gullies on both sides of a north-south country road 1226 m north of farm to market road 1949, and 3218 m north of Enloe, Delta County, Cooper North quadrangle.

8-10-911 (15) Pecan Gap Chalk. In stream bed just south of farm to market road 2456, 4.02 km east-southeast of Ladonia, Fannin County, Ladonia quadrangle.

10-9-7-4a (16) Rockwall Member of the Pecan Gap Chalk. In a ditch on the east side of dirt road leading up hill to the south of farm road 740, 1000 m north of U.S. Interstate 30, Rockwall County, Rockwall quadrangle.

8-8-9-1.d (10) Wolfe City Sandstone. In cliff on North Bank of north Sulphur River, just east of Texas Highway 19 and 24 on the Delta County - Lamar County line, about 20.5 km south of Paris, Cooper North quadrangle.

8-9-9-7 (11) Wolfe City Sandstone. Bluff on North Sulfur River at farm to market road 2675, 8.2 km south-southeast of Roxton, Lamar County, Cooper North quadrangle.

10-9-7-3f (12) Brownstown Marl. Santa Fe Railroad cut, 30 m north of interstate 30 and intersecting farm road 740 near Lake Ray Hubbard, Rockwall County, Rockwall quadrangle.

10-9-7-3e (13) Same as above (see Thompson 1983 for diagram showing the 10-9-73 locality and collecting sites).

7-22-9-3i (05) Annona Chalk. Ditch by farm to market road 1158, 11.1 km east-northeast of Clarksville, Red River County, Whiterock quadrangle.

7-21-9-81 (06) Annona Chalk. Ditch by farm to market road 1158, at north-south to east-west curve, 8.0 km north-northwest of Annona and 11.9 km east-northeast of Clarksville, Red River County, Whiterock quadrangle.

7-22-9-3e (09) Same as 7-22-9-3i

7-22-9-3c (08) Same as 7-22-9-3i

7-22-9-3b (07) Same as 7-22-9-3i

8-8-9-5 (98) Gober Chalk, (in back of tank) just north of county road 1.6 km east of U.S. 271, 4.66 km northwest of Pattonville, 13.5 km southeast of Paris, Lamar County, Pattonville quadrangle.

8-12-9-6 (00) Gober Chalk in Brally Pool creek and county road 3.38 km east of farm to market road 1743, 9.0 km southwest of Honey Grove, Fannin County, Honey Grove quadrandle.

11-3-8-3 (97) Gober Chalk. In bed of Bailey creek just south of Texas highway 78, 8.84 km west of Leonard, Fannin County, Trenton quadrangle.

8-9-9-9 (99) Gober Chalk. In creek bed on east side of county road 3.62 km southeast of Petty and 6.67 km northwest of Roxton, Lamar County, Honey Grove quadrangle.

8-9-9-3 (02) Gober Chalk. In stream bed of Mallory creek on farm to market road 2122, 6.43 km northeast of Roston and 13.8 km southwest of Paris, Lamar County, Paris quadrangle.

7-1-9-2 (01) Gober Chalk. Hillside on east side of snow hill road (west of state highway 78, and north of farm to market road 1553) 6.75 km east-northeast of Randolph and 3.8 km north of farm to market road 1553, Fannin County, Bonham quadrangle.

8-12-9-3 (03) Gober Chalk. In the bed of Bullard creek and county road, 3.6 km southeast of farm to market road 1743 and 9.0 km south of Windom, Fannin County, Honey Grove quadrangle.

11-3-8-4d (04) Gober Chalk. Road cut just north of Lindsey creek on farm to market road 151, 1.2 km north of Texas highway 11, and 2.25 km east-southeast of Whitewright, Fannin County, Whitewright quandrangle.

9-24-7-9 (90) Brownstown Marl. 4.02 m west of bridge on farm road 2990 in cliffs on the North Sulphur river about 3.2 km north-northwest of Ladonia, Fannin County, Ladonia quadrangle.

9-15-9-10 (91) Brownstown Marl. In a creek bed just northeast of county road, 1.8 km west of U.S. highway 69, and 5.95 km north-northwest of Celeste, Hunt County, Celeste quadrangle.

8-8-9-8 (92) Brownstorn Marl. In stream bed at farm to market road 137, 9.65 km west of Howland, 15.9 km south-southwest of Paris, Lamar County, Paris quadrangle.

7/1-9.5b (94) Brownstown Marl. In deep stream cut just south of U.S. Highway 82, 6.43 km east of Bonham town center, 33.0 m west of "rest stop", Fannin County, Bonham quadrangle.

8-9-9-18 (95) Brownstown Marl. In gully just west of county road 1.6 km south of Cottonwood Creek, 2.7 km northwest of Windom, and 5.6 km northeast of Dodd City, Fannin County, Bonham quadrangle.

8-27-7-2d (88) Austin Chalk. In creek bed about 4.02 km south of Murphy road, and 42.8 km northwest of Texas highway 78, 1.4 km west southwest of town center of Sachse, Dallas County, Rowlett quadrangle.

8-21-6-3b (89) Austin Chalk. 30m north of June Lake road on Praris Creek, Dallas County, Whiterock Lake quadrangle.

Appendix B

Sample Processing Technique

Hand Method

1. Wash sample if consolidated. Scrape off any porous material surrounding concretions.
2. Test the sample to determine whether or not it is calcareous.
3. If the sample is not calcareous, put a piece approximately 3cm square in a pie tin, cover it with another pie tin, and break it up with a hammer into 1cm size pieces. Wipe the hammer and the anvil with a damp sponge between samples.
4. If the sample is calcareous use more material and break it up more finely.
5. Place each non-calcareous and slightly calcareous sample in a plastic cup. Label each cup with the sample number.
6. Place very calcareous samples in large glass beakers and label the beakers with the corresponding sample numbers.

HCl Treatment (For Calcareous Samples)

1. Dampen the sample with water.
2. Slowly add concentrated HCl and let the sample stand. If the sample starts to overflow the beaker, add a few drops of Acetone to break up the foam.

3. Stir occasionally and add more concentrated HCl periodically until additional HCl does not cause any fizzing. Now the carbonates are dissolved.

4. Put the sample into one or more labeled, 50 ml, plastic test tubes. Wash all fines from the sample into the test tubes. Any remaining large chunks can be rinsed 2 or 3 times in the beaker with momentary settling and decanting into the laboratory sink. The test tubes containing the fines must be centrifuged (see below) and decanted.

5. Rinse the sample in centrifuge tube at least twice. Transfer coarse material to labeled plastic cup.

6. Settling can be used instead of centrifuging if desired. Fill the beaker containing the sample and HCl, with water. Allow to settle for at least two hours.

Centrifugation and Washing

1. Fill the centrifuge tube. Centrifuge the sample and decant the supernatant.

2. After the first centrifugation, the sample will be compressed at the bottom of the tube. If there is more than 2cm of sample in the test tube at this time, the sample will not wash well; put some of the sample into another labeled tube.

3. Centrifuge until supernatant liquid is clear.

4. Repeat centrifugation and washing until the

HF Treatment

1. Make sure you are wearing a lab coat, gloves, and a face shield. Always work under the ventilated fume hood. Have a large plastic container with some water sitting in the hood sink.

2. Label samples in 50 ml plastic tubes (coarse fractions and non-calcareous samples). Label a plastic cup for any sample that does not have one. Dampen with water any sample that is dry. Have a spare clean cup close by the labeled one for extra capacity if needed in step 3.

3. SLOWLY add HF (ca. 50%) to the fine fraction in the plastic tube, covering it with 3cm of acid. CAREFULLY stir with a stirring rod, holding the tube at a slant and over the appropriate plastic cup. The reaction may or may not start at once, and may start to foam and overflow the tube quite suddenly. As the reaction proceeds, dump the acid and fine fraction into the plastic cup. Add more acid by rinsing the tube and/or adding to the cup until the sample is covered with about 2cm of acid. Swirl the plastic cup continuously to agitate. Watch the reaction carefully for the first few minutes; add more HF if necessary. If sample foams beyond capacity of

labeled cup, let it flow into spare cup. An uncontrollable reaction can be partially dumped in a large container under the hood. If no vigorous reaction has taken place, keep an eye on the sample for at least 5 minutes after adding the last HF, since the reaction may be delayed.

4. Let samples remain in HF until all visible reactions have ceased (usually 10 to 15 minutes). Then transfer to labeled plastic 50 ml plastic tubes, leaving undissolved rock fragments in the plastic cup, but washing all fine materials into the tube(s).

5. Centrifuge sample and decant into a plastic container in the hood sink. Neutralize the acid. Rinse sample at least 3 times, then transfer to 50 ml glass tube(s).

6. Rinse out the plastic cups that contained the HF, leaving rock fragments in the cups. Let dry thoroughly in the hood before you discard them.

Hot HCl Treatment

1. Add to (50 ml) glass centrifuge tube containing residue an amount of concentrated HCl equal to or slightly more than the volume of the residue.

2. Stirring continuously with glass stirring rod, bring to a boil under the fume hood, (about 1-2 minutes).

3. Let the tube cool briefly, then fill with water. Centrifuge and decant into container in hood sink, but do not wash the residue.

"Clorox" Treatment

1. Label a medium sized beaker (400 ml or so) with the sample number. Keep this handy in case the reaction foams over the tube.
2. Slowly add approximately 20 ml of "Clorox" (or any 5-6% Sodium Hypochlorite solution) to the 50 ml glass test tube containing the sample. Stir with a stirring rod and hold at a slant over the labeled beaker (be sure to stir loose all the residue that may be packed against the glass tube). If reaction is violent, dump mixture into beaker. If it is quiet (and residue is originally light-colored, or of a small volume if dark) reaction can proceed in centrifuge tube. Be alert to delayed reaction.
3. Allow the mixture to react 10 to 20 minutes, stirring occasionally. Watch for color change as an indication of reaction. If there is a large amount of organic matter in the sample, test to see if there is sufficient "Clorox" by adding concentrated HCl drops - fizzing means sufficient. Be sure to end the treatment with an excess of "Clorox". Use the labeled

beaker any time you need more space. Reaction in obviously organic-rich samples which are sluggish can be encouraged by alternate addition of "Clorox" and HCl in quantities sufficient to shift the solution substantially over the pH 7 point.

4. Centrifuge and decant. Rinse once.

"Labtone" Treatment

You will get a better separation of organic and mineral materials with the heavy liquid treatment, if you can discard some of the fine mineral particles now. A short centrifugation with a solution of Labtone (a laboratory detergent) will keep these particles in suspension is that they can be discarded.

For Labtone solution:

-600 ml warm water

-16 gm Labtone

Shake well to dissolve the Labtone, and sieve through a 35 micron mesh. The solution will turn cloudy when it cools but that does not affect its performance.

1. If centrifuged residue occupies more than one inch of 50 ml tube, split it into two or more fractions, as needed.
2. Add a few ml of the Labtone solution to each tube (in 50 ml tube).
3. Mix sample well.

4. Fill each tube 1/4 full with the Labtone solution and then fill with filtered water.

5. Centrifuge for 1 minute and gently decant supernatant.

6. Fill again with filtered water, and centrifuge for 50 seconds, gently decant; repeat centrifuge for 40 seconds, then 35 seconds. Repeat 35 seconds until supernatant liquid is clear. (If in doubt about throwing away fossils, check some supernatant under the microscope.) This treatment can be repeated, or centrifuging time reduced still more (to 30 seconds) if it seems appropriate.

7. Centrifuge and rinse until free of all detergent, ending with a 3 minute centrifugation to pack residue firmly. Decant as thoroughly as possible.

Preparing for the Heavy Liquid Separation

You will need a solution of $ZnBr_2$ with a specific gravity of 2.0:

- 412 ml concentrated $ZnBr_2$ (500, or 1500 ml)
- 240 ml water (291, or 873 ml)

Mix the $ZnBr_2$ and water THOROUGHLY (pouring forth and back between two containers is satisfactory, but simple stirring is not adequate). Check the solution with the hydrometer. Add more $ZnBr_2$ or water until the specific gravity is 2.

You will also need to make Bostick tubes, as follows:

1. For each tube, cut a piece of Tygon tubing 1.7cm (5/8 inch i.d.) 9cm long.
2. Place the tube in a beaker of very hot water for a few seconds to allow it to regain a cylindrical shape.
3. With a file, mark a shallow cross in the wider end of a #2 rubber cork. This is to prevent the cork from sticking to the bottom of the centrifuge tube after centrifugation. Insert the narrow end of the cork as far as possible into one end of the Tygon tube.
4. Using a pair of pliers, bend off the CIRCULAR ring from a pull tab from an aluminum can (beer, soft drink, etc.). Slide the ring over the tube to the tube-cork junction and then force it further over the cork to seal the tube tightly to the cork. A washer of the same size 1.7cm (5/8" inner diameter) may be used instead of the pull tab ring. A short piece of thick walled metal tubing that just fits over the Tygon tube, can be used to help push the ring into place.

Heavy Liquid Separation

1. Samples have been centrifuged (3 minutes) and thoroughly rinsed at the end of the last step.
2. Add approximately 10 ml of $ZnBr_2$ (sp. gr. = 2.0) to each sample, and mix thoroughly.

3. Transfer each sample mixed with $ZnBr_2$ from the 50 ml tube to a labeled Bostick tube in one hand with the open end closed securely by a finger protected with a rubber fingercot, invert the tube several times to thoroughly mix the contents. The mixture should be completely homogeneous; any lumps should be dispersed with a stirring rod before proceeding. The fingercot need not be replaced when treating a succession of samples, if the surface is washed carefully under running water to avoid contamination.

4. Place the Bostick tube in a clear labeled 50 ml plastic tube. Fill the outer tube with water to a level just above that of the $ZnBr_2$ in the Bostick tube, being careful not to get any water into the $ZnBr_2$ suspension.

5. Centrifuge for 20 minutes.

6. The organic materials in the sample are now floating on top of the $ZnBr_2$. These organic material must now be transferred to a 15 ml labeled glass tube, in the following way: with long-nosed pliers, firmly pinch the Bostick tube well below the "float" and GENTLY pour the material above the pinch into the 15 ml glass tube; flush out with water jet from squeeze-bottle, being sure to retain firm pressure on the pliers. Add a drop of concentrated HCl to the 15

ml tube to prevent the formation of $Zn(OH)_2$ (a flocky white precipitate which is soluble in HCl). Add water to the 15 ml tube, invert to MIX WELL, and centrifuge for 90 seconds.

7. Decant, refill with water and centrifuge 1 minute. Repeat.

If the "float" layer on the $ZnBr_2$ is more than 3 mm thick, divide the sample between two tubes for rinsing to keep the specific gravity low enough to allow the organic material to settle easily. Add HCl to each tube.

Acetolysis Treatment

1. Centrifuge and wash the samples (15 ml glass tubes labeled with both: Magic Marker and Wax Pencil).
2. Fill each tube about half full with Glacial Acetic Acid (HAc).
3. Centrifuge and decant the HAc, DO NOT add water.
4. Fill each residue-bearing tube about half full with Acetic Anhydride.
5. Stir each sample with dry stirring rod. Leave the stirring rods in the centrifuge tubes.
6. CAUTIOUSLY add about 10 drops of concentrated H_2SO_4 to each sample. Stir.
7. Place the centrifuge tubes in a warm water bath in the fume hood (a 250 ml beaker will hold eight 15 ml tubes). Stir every few minutes as you bring the

water to boiling and keep it boiling gently for about 10-15 minutes, until the Acetolysis mixture turns a very deep tea color.

8. Remove tubes from water bath to test tube rack and, as soon as you can handle them, centrifuge for 2 minutes. CAREFULLY decant the supernatant fluid into a beaker full of cold water (unless there is a large excess of water, the fluid will pop and sputter).

9. Add a few (10) ml of Glacial Acetic Acid (HAc) to each sample.

10. Centrifuge and wash with water 3 times. If the residue is not to be processed further at once, add a few (2 droppers-full) Phenol (5%) to the water covering the residue. Renew the labels of tubes as necessary.

Preparing for the Sieve Separation

You will need two sieves of mesh: 35 micron, and 15 micron. Brass frame sieves can be used, but involve cleaning problems, or expendable sieves can be prepared in the following way:

1. Cut the top off four plastic 15 ml test tubes about 5cm from the top, and smooth off the cut edges.

2. Mark each cut top with a letter A or B , one each.

3. Cut a 12cm x 20cm rectangle of nylon monofilament bolting cloth of 35 micron grade. Fold a piece of

21.5cm x 28cm (8½ x 11") paper in half and insert the mesh. Cut with a paper cutter into 2.5cm squares, using the surrounding paper to avoid touching the bolting with the fingers, and cut it more easily. Store the cut squares in a dry plastic container. Repeat for 15 micron squares. Use a snugly fitting rubber hose washer to fix the appropriate mesh square onto the cut end of the plastic tube.

4. After sieving, the nylon screen should be discarded and the tube and washer washed thoroughly.

Sieve Separation

1. Stir the residue in the 15 ml tube.
2. Label two 50 ml beakers with the sample number, and label two watch glasses with the same number followed by A or B.
3. Hold the sieve over one beaker and pour the residue through. Rinse out the centrifuge tube onto the sieve.
4. Rinse the residue through the sieve by directing a squirt of water from squirt bottle through residue and sieve. An eye dropper can be used to agitate the residue if it clogs the sieve.
5. Add a dropper of Darvan (5%) to residue and rinse through sieve if residue seems at all clumped or difficult to rinse. This is usually necessary on the "B" sieve.

6. Invert the "A" sieve onto the "A" watch glass and rinse residue out by a squirt of water on the bottom of the sieve. Add a dropper-full of Phenol to avoid fungal attack to the sample.

7. Repeat for Sieve "B".

Swirling

This technique was used to separate residues according to the size and composition of particles before the development of heavy liquid and sieving techniques. For those purposes it is effective, but excessively time-consuming. However, it is still the best technique for concentrating small amounts of residue into a small volume immediately before preparing slides.

The procedure is as follows:

1. Place a watch glass containing water and a small amount of residue on a smooth surface and allow the suspension to settle for a minute or two.

2. Grasp the watch glass firmly between thumb and forefinger, rest the heel of your hand on the table, keep the watch glass in contact with the table, and keep the rim of the watch glass at level. With your arm, not fingers, move the watch glass smoothly in circles of 2-4 mm diameter. The movement can be rapid, but the object is to avoid turbulence and sloshing the liquid. Fossils and debris will move

gradually toward the center of the watch glass and will arrange themselves there according to their differences in density and surface area.

By this method, even a few specimens of a poorly fossiliferous sample can be concentrated from several mls of water and located for mounting or individual handling.

Clumping (electrostatic) is sometimes a problem at this stage. Experiment with different kinds of water (tap, distilled, deionized) or add a drop of ammonia to the swirl dish.

Glycerine - Jelly Slides

1. Turn on the warming table (at about 50°C) or hot plate (200°F), and place the dropping bottle of glycerine jelly on the warming table.
2. Use the glass-marking scribe to number each slide toward its upper left hand corner with the sample number, and in the upper right corner with the fraction code. Plan on making at least one slide for each fraction containing fossils and two (or more) for each good fraction of exceptionally good samples.
3. Place the numbered slides on the table and place the coverslips next to the corresponding slide.
4. The residue should be in a watch glass. Swirl to gather the fossils together.

5. Draw up a few drops of residue in an unused disposable pippette and allow a bubble to rise through the suspension in the pippette to homogenize it. Then drop a few drops on the coverslip. Use a new pippette for each sample.

6. Add a few drops (2-3) of melted glycerine-jelly (or a crumb of solid, cool jelly) to the coverslip and mix well with the residue using the stirring rod (a clean one for each sample). Spread the residue and glycerine mixture as evenly as possible, almost to the edge of the coverslip.

7. Allow the coverslip to dry undisturbed on the warming table. To see if it is dry, hold a cool slide over it, touching the warming table just next to one edge of the coverslip, but not allowing the strew to touch the slide. If no condensation appears on the slide, the strew is dry.

8. Remove the coverslip (dry) from the warming table and let it cool completely. Slightly warm the corresponding numbered slide and place it in the slide holder. Immediately add a few drops of the warm glycerine jelly, with the stirring rod from the dropping bottle.

9. Promptly but gently invert the coverslip onto the slide, lowering it at one end first, with care to

prevent trapping of air bubbles. Quickly press the coverslip down with a pair of forceps so that the glycerine jelly mixture flows beyond the edges of the coverslip. Press out any bubbles.

10. Remove the slide from the slide holder and place it in a slide tray. Allow the slide to dry 3-4 days before you clean, number, and seal it.

Cleaning Glycerine-Jelly Slides

Glycerine-jelly slides must be cleaned and sealed before they are numbered and stored:

1. Cut or scrape away most of the excess glycerine jelly around the cover slip.
2. Use a Kimwipe dipped in hot water (not boiling) to wipe away the remaining glycerine jelly. Keep a small amount of hot water in an aluminum pie pan over Bunsen burner. Dry the slide with a dry Kimwipe. Leave no smears of glycerine jelly at the coverslip edges.
3. Seal the edges of the coverslip with clear nail polish, using a fine brush or dropper.

Storing Residues

Residues can be stored in water and Phenol (or alternately 95% ethyl alcohol). To store in water and Phenol:

1. Transfer unmounted residue from watch glass (with an unused pippette) or from centrifuge tube into a 1½ dram vial.
2. Add filtered water to about 3/4 full.
3. Add 1 dropper-full of Phenol and 2-3 drops of glycerine.
4. Cover the top of the vial with a 2.5cm square inch piece of Parafilm, and screw on the cap firmly.
5. Use a gummed label (not pressure sensitive!) or portion of one to label the vial with the sample number. Also record the Processing Sheet number. Use a circular pressure sensitive label to label the cap with the sample number.

Appendix C

Plates

PLATE 1

1. Ascodinium acrophorum Slide no. 507, h. 7.9, v. 9.9. 72 x 52 Microns.

2. Subtilisphaera sp. Slide no. 503, h. 10.8, v. 18.0 70 x 40 Microns.

3. Alterbia acutula Slide no. 497, h. 23.4, v. 14.5. 100 x 62 Microns.

4. Alterbia sp. C. Slide no. 504, h. 20, v. 11. 80 x 52 Microns.

5. Alterbia sp. A. Slide no. 513, h. 23.2, v. 17.2. 60 x 40 Microns.

6. Laciniadinium arcticum Slide no. 518, h. 5.9, v. 11.8. 42 x 40 Microns.

7. Alterbia sp. B. Slide no. 505, h. 15.5, v. 11.2. 120 x 74 Microns.

8. Alterbia sp. B. Slide no. 514, h. 17.3, v. 12.0. 138 x 80 Microns.

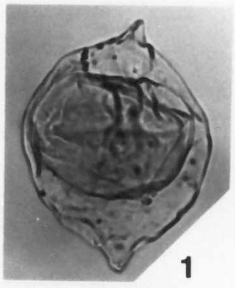
9. Ceratiopsis pannucea, Slide no. 490, h. 21.7, v. 7.7. 150 x 94 Microns.

10. Phelodinium tricuspis, Slide no. 478, h. 27.4, v. 11. 140 x 90 Microns.

11. Ceratiopsis diebelii, Slide no. 513, h. 26.2, v. 17. 120 x 80 Microns.

12. Ceratiopsis pannucea, Slide no. 492, h. 8.6, v. 13. 175 x 90 Microns.

PLATE I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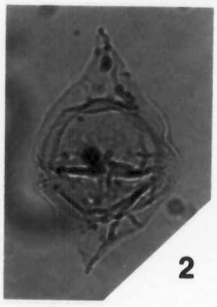
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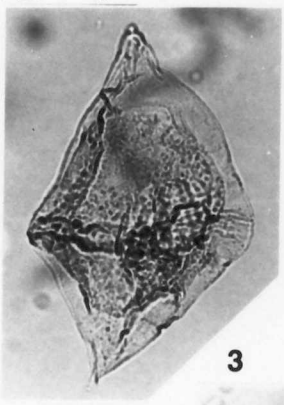
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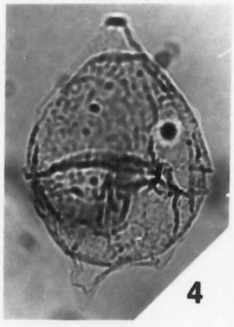
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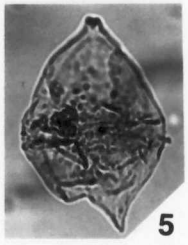
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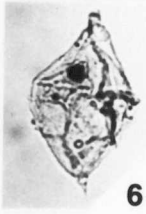
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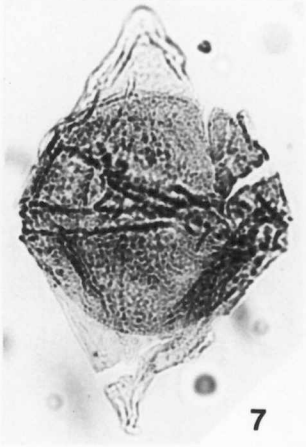
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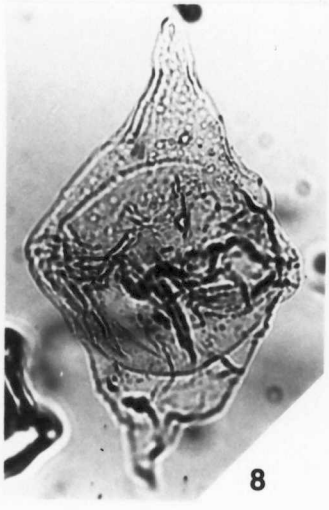
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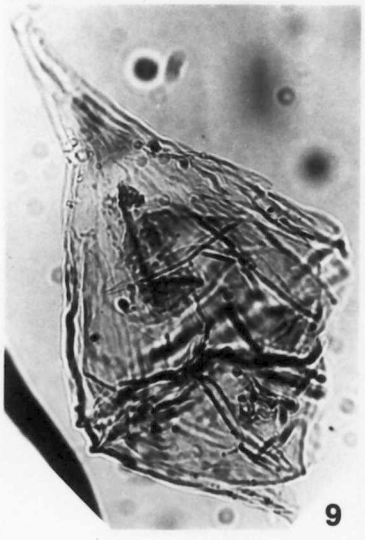
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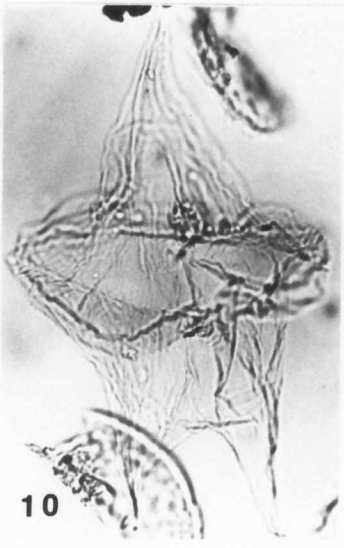
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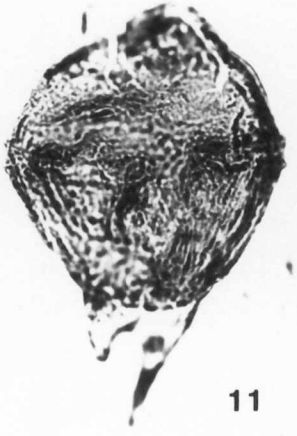
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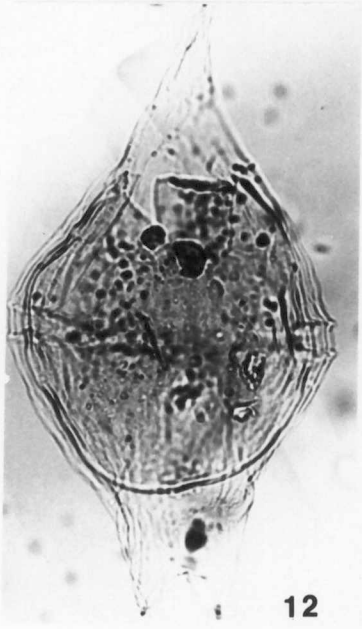
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PLATE 2

1. Lejeunecysta sp. B. Slide no. 523, h. 8.8, v. 18.2. 65 x 26 Microns.

2. Lejeunecysta sp. B Slide no. 523, h. 12.1, v. 7.6. 50 x 50 Microns.

3. Lejuenecysta sp. A Slide no. 495, h. 10.3, v. 2.2. 90 x 66 Microns.

4. Rottnestia borussica Slide no. 503, h. 10.0, v. 9.5. 82 x 58 Microns.

5. Chatangiella victoriensis Slide no. 489, h. 3.9, v. 43.2. 100 x 60 Microns.

6. Chatangiella granulifera Slide no. 113, h. 13.2, v. 12.0. 82 x 52 Microns.

7. Chatangiella tripartia Slide no. 590, h. 6.0, v. 18.2. 76 x 40 Microns.

8. Unknkown Deflandre Slide no. 523, h. 17.2, v. 9.3. 80 x 60 Microns.

9. Palaeocystodinium sp. A Slide no. 514, h. 7.5, v. 3.2. 120 x 50 Microns.

10. Palaeocystodinium sp. C Slide no. 513, h. 10.0, v. 14.2. 150 x 60 Microns.

11. Chatangiella granulifera Slide no. 489, h. 10.0, v. 27.4. 90 x 55 Microns.

12. Deflandre sp. A Slide no. 423, h. 28.1, v. 5.9. 90 x 70 Microns.

13. Palaeoperidinium sp. A Slide no. 491, h. 15.6, v. 16.1. 100 x 95 Microns.

14. Deflandre sp. A Slide no. 423, h. 27.2, v. 8.7. 90 x 75 Microns.

15. Deflandre sp. B Slide no. 423, h. 29.3, v. 8.7. 80 x 104 Microns.

PLATE 2


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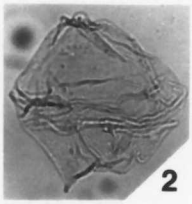
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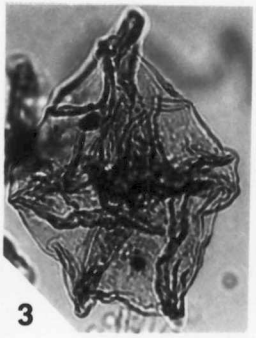
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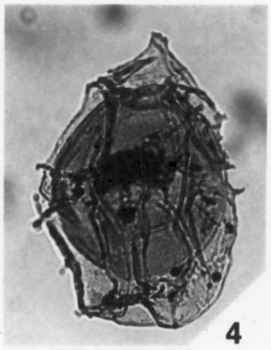
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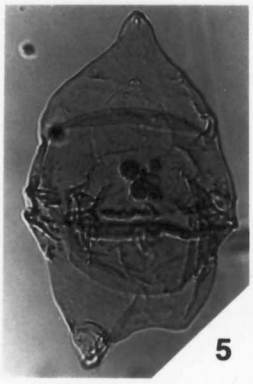
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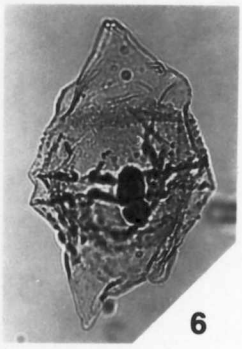
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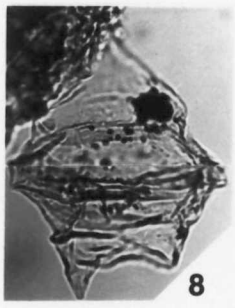
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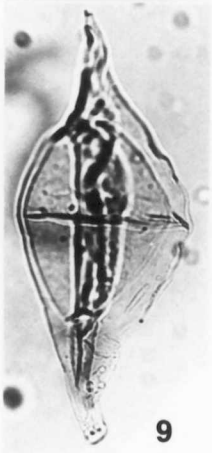
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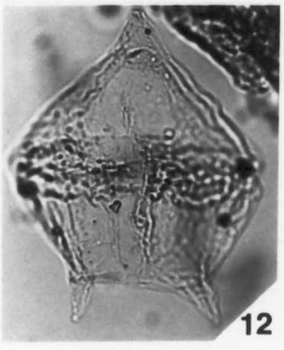
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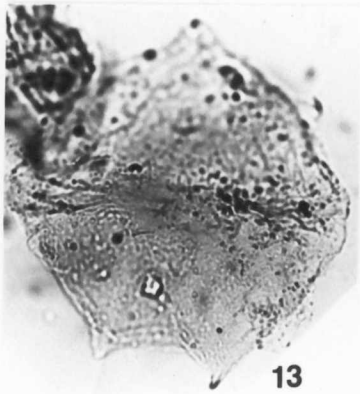
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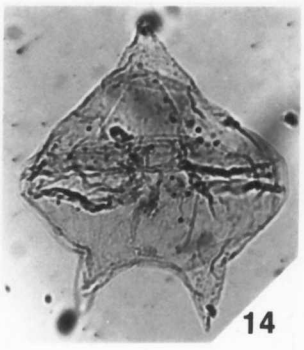
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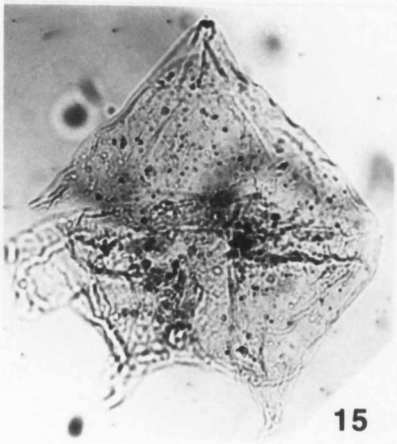
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PLATE 3

1. *Spinidinium* sp. B Slide no. 518, h. 7.2, v. 16.4. 56 x 44 Microns.

2. *Spindinium* sp. G Slide no. 523, h. 27.5, v. 10.5. 60 x 40 Microns.

3. *Spinidinium densispinatum* Slide no. 502, h. 15, v. 6.6. 64 x 50 Microns.

4. *Spinidinium essoi* Slide no. 502, h. 14, v. 7.1. 70 x 58 Microns.

5. *Spinidinium sverdrupianum* Slide no. 509, h. 26.8, v. 18.3. 70 x 48 Microns.

6. *Spinidinium* sp. B Slide no. 518, h. 17.7, v. 16.6 53 x 43 Microns.

7. Unknkown Peridinoid Slide no. 518, h. 19.8, v. 6.1. 60 x 44 Microns.

8. *Paleohystrichophora infusorioides* Slide no. 491, h. 12.7, v. 13.2. 70 x 50 Microns.

9. *Spinidinium sverdrupianum* Slide no. 505, h. 9.8, v. 14. 80 x 56 Microns.

10. *Palaeohystrichophora* sp. B Slide no. 511, h. 7.8, v. 10.2. 110 x 58 Microns.

11. *Palaeohystrichophora* sp. C Slide no. 90, h. 11.2, v. 9.6. 84 x 44 Microns.

12. *Palaeohystrichophora infusorioides* Slide no. 491, h. 12.0, v. 7.7. 60 x 40 Microns.

13. *Spinidinium echionoideum* Slide no. 498, h. 11.7, v. 10.1. 86 x 70 Microns.

14. *Isabeladinium* sp. A Slide no. 523, h. 11.6, v. 7.7. 65 x 58 Microns.

15. *Isabelladinium acuminatum* Slide no. 515, h. 36.2, v. 10.0. 82 x 48 Microns.

16. *Trithyrodinium* sp. C Slide no. 513, h. 21.5, v. 5.2. 72 x 55 Microns.

17. Isabelidinium belfastense Slide no. 491, h. 7.7, v. 11.2. 98 x 56 Microns.

18. Isabelidinium microarmum Slide no. 502, h. 21.1, v. 17.6. 90 x 62 Microns.

19. Trithyrodinium sp. B Slide no. 498, h. 11.6, v. 7. 82 x 70 Microns.

20. Trithyrodinium sp. A Slide no. 409, h. 6.5, v. 10.2. 115 x 80 Microns.

PLATE 3

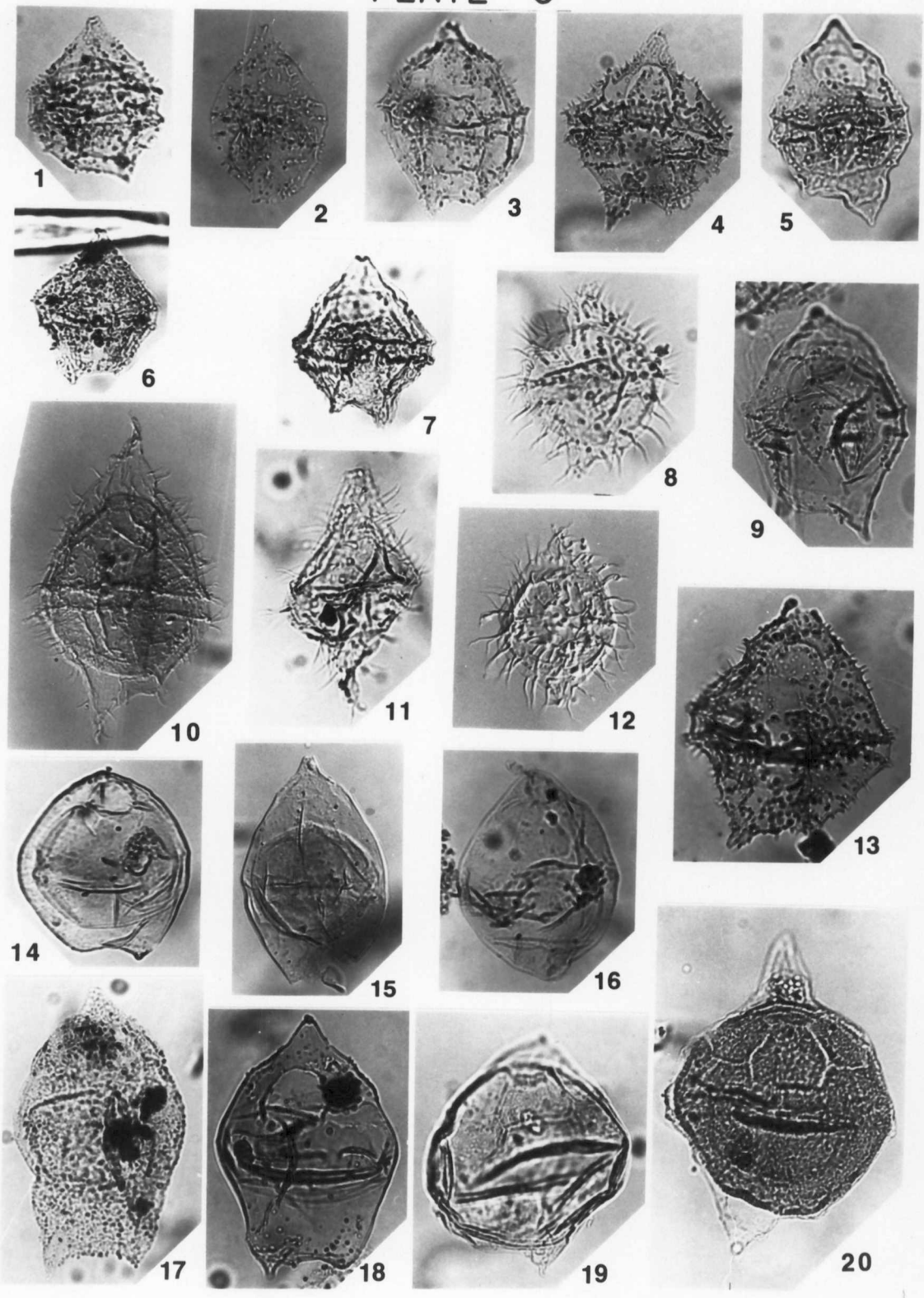


PLATE 4

1. Unknown Peridinioid Slide no. 518, h. 14.7, v. 7.3. 74 x 53 Microns.

2. Unknown Peridinioid Slide no. 518, h. 8.9, v. 3.1. 74 x 53 Microns.

3. Spinidinium clavus Slide no. 490, h. 201, v. 6.0. 58 x 44 Microns.

4. Spinidinium clavus Slide no. 490, h. 15.8, v. 5.1. 54 x 43 Microns.

5. Hystrichosphaeridium palmatum Slide no. 498, h. 18.9, v. 6.2. 28 Microns Inner Cyst.

6. Hystrichosphaeridium tubiferum subsp. brevispinum Slide no. 512, h. 7.0, v. 11.2. 64 x 52 Microns Inner Cyst.

7. Hystrichosphaeridium sp. X Slide no. 523, h. 8.1, v. 4.7. 36 Microns Inner Cyst.

8. Hystrichosphaeridium tubiferum Slide no. 504, h. 26.2, v. 12.6. 50 Microns Inner Cyst.

9. Hystrichosphaeridium sp. A Slide no. 501, h. 8, v. 10.4. 40 x 48 Microns Inner Cyst.

10. Cyclonephelium distinctum Slide no. 497, h. 25.7, v. 6.7. 60 x 80 Microns Inner Cyst.

11. Hystrichokolpoma rigaudae Slide no. 512, h. 12.0, v. 7.7. 54 Microns Across Cingular Area.

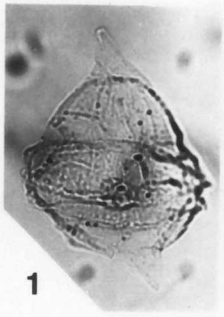
12. Cyclonephelium sp. C Slide no. 521, h. 10.7, v. 7.7. 64. x 80 Microns.

13. Diphyes sp. A Slide no. 501, h. 13.8, v. 7.9. 42 x 40 Microns Inner Cyst.

14. Cyclonephelium compactum Slide no. 490, h. 4.3, v. 10.2. 90 Microns Across Cingular Area.

15. Cyclonephelium distinctum Slide no. 521, h. 9.2, v. 9.3. 100 x 100 Microns Inner Cyst.

PLATE 4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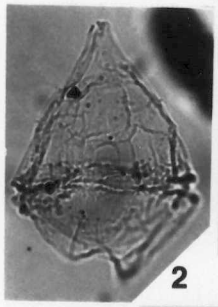
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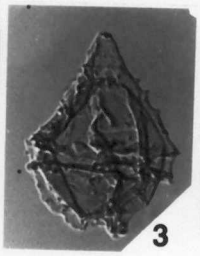
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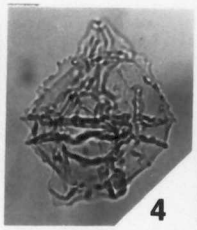
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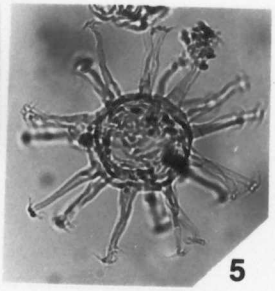
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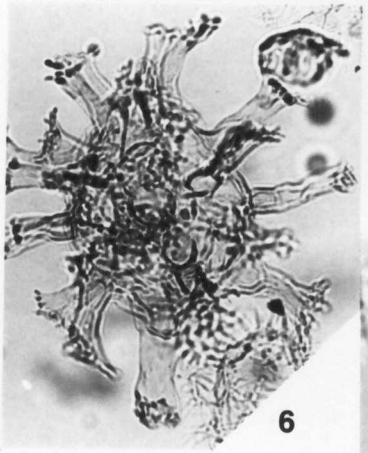
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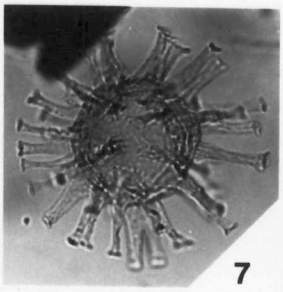
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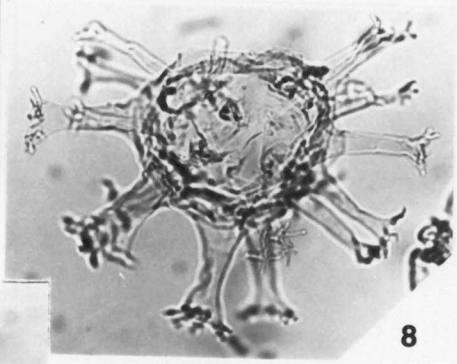
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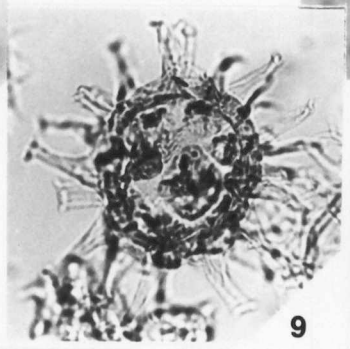
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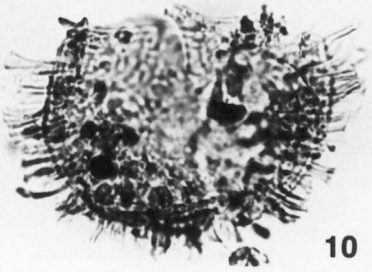
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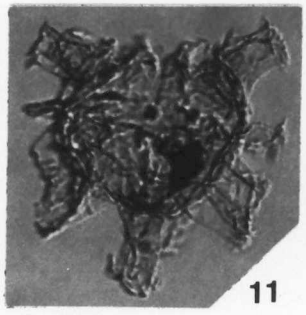
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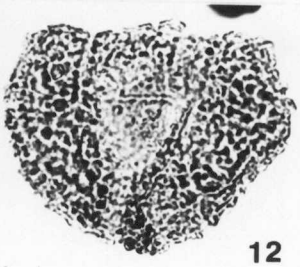
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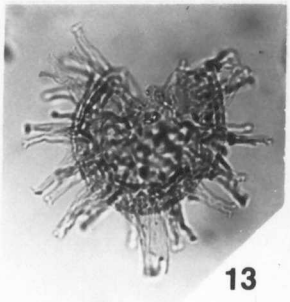
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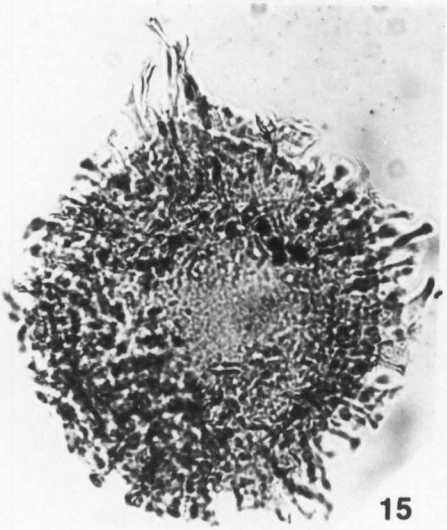
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PLATE 5

1. Spiniferities granulatus Slide no. 501, h. 9.6, v. 1.9. 64 x 56 Microns Inner Cyst.

2. Spiniferities granulatus Slide no. 491, h. 5.5, v. 10.6. 40 Microns Inner Cyst.

3. spiniferites cingulatus Slide no. 501, h. 13.8, v. 3.2. 60 Microns Inner Cyst.

4. Florentinia laciniata Slide no. 497, h. 23.9, v. 3.7. 60 Microns Inner Cyst.

5. Spiniferities ramosus Slide no. 497, h. 28.6, v. 4.8. 67 x 52 Microns Inner Cyst.

6. Spiniferities ramosus Slide no. 491, h. 12.7, v. 10.4. 54 x 44 Microns Inner Cyst.

7. Florentinia sp. A Slide no. 497, h. 9.4, v. 9.2. 48 x 42 Microns Inner Cyst.

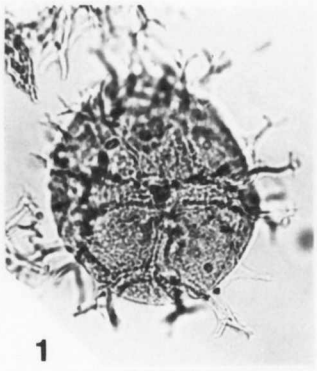
8. Florentinia sp. B Slide no. 494, h. 13.9, v. 14.8. 46 Microns Inner Cyst.

9. Florentinia clavigera Slide no. 504, h. 13.8, v. 4.1. Overall 149 Microns.

10. Eatonicysta sp. A Slide no. 498, h. 36.5, v. 24.7. 120 x 100 Microns.

11. Eatonicysta sp. A Slide no. 500, h. 20.2, v. 16.0. 130 x 108 Microns.

PLATE 5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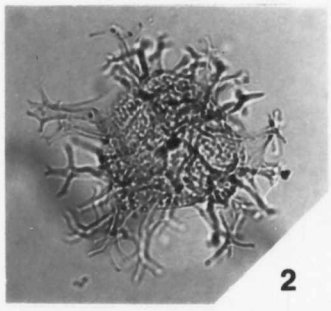
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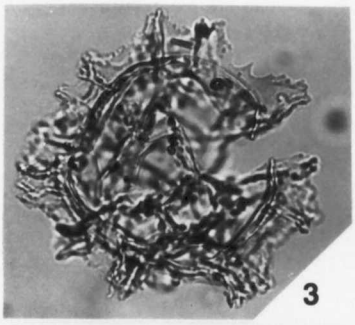
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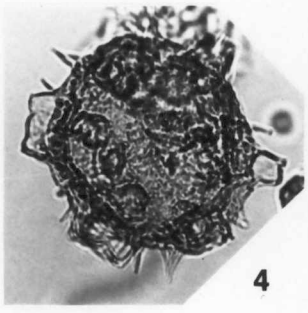
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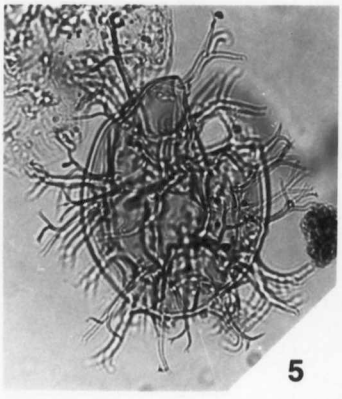
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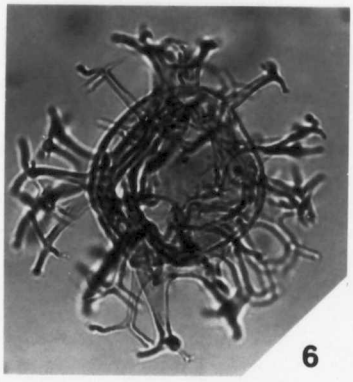
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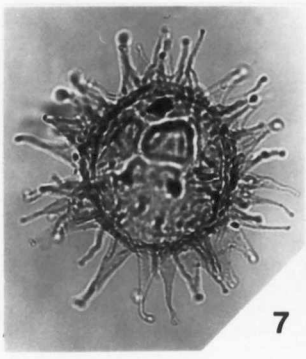
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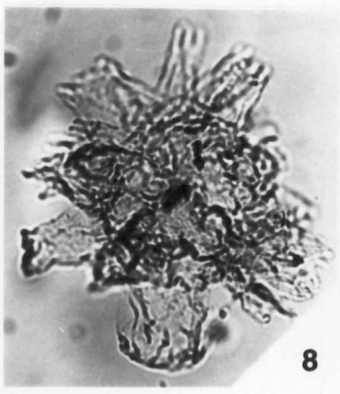
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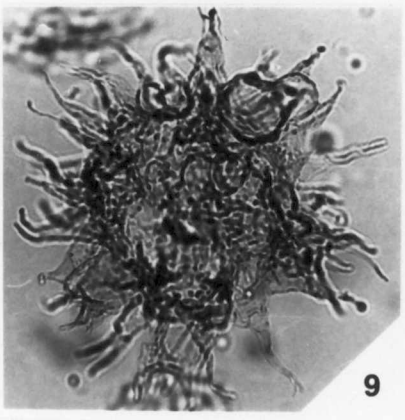
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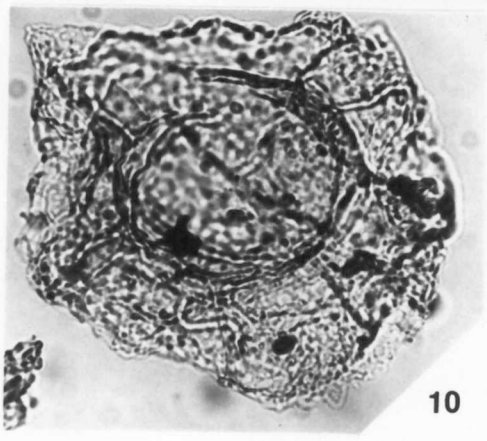
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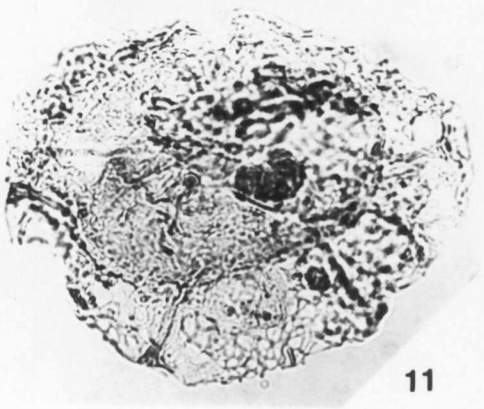
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PLATE 6

1. Tanyosphaeridium xanthiopyxides Slide no. 498, h. 28.8, v. 5.7. 40 x 22 Microns.

2. Tanyosphaeridium xanthiopyxides Slide no. 503, h. 24.2, v. 4.4. 44 x 22 Microns.

3. Aiora sp. Slide no. 508, h. 18.5, v. 11.9. 60 Microns Overall.

4. Areoligera senonensis Slide no. 506, h. 40.9, v. 11.8. 64 Microns Inner Cyst.

5. Senoniasphaera rotundata Slide no. 503, h. 14.2, v. 11.5. 70 Microns Wide Inner Cyst.

6. Senoniasphaera protrusa Slide no. 491, h. 21.2, v. 11.3. 70 Microns Inner Cyst.

7. Unknown sp. A Slide no. 523, h. 7.4, v. 1.4. 140 x 90 Microns.

8. Senoniasphaera protrusa Slide no. 504, h. 6.5, v. 14.3. 84 x 70 Microns Inner Cyst.

9. Unknown sp. 506 Slide no. 506, h. 44.5, v. 17.5. 80 x 54 Microns.

10. Nematosphaeropsis pusulosa Slide no. 501, h. 19.8, v. 7.9. 60 Microns Overall.

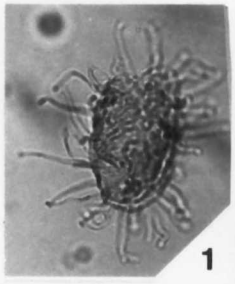
11. Adnatosphaeridium vittatum Slide no. 506, h. 27.1, v. 4.8. 70 Microns Inner Cyst.

12. Senoniasphaera rotundata Slide no. 503, h. 12.9, v. 12.5. 68 Microns Inner Cyst.

13. Nematosphaeropsis pusulosa Slide no. 498, h. 10.3, v. 11.2. 68 Microns Inner Cyst.

14. Adnatosphaeridium vittatum Slide no. 498, h. 24.7, v. 7.9. 60 Microns Inner Cyst.

PLATE 6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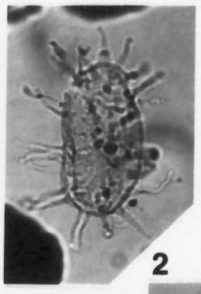
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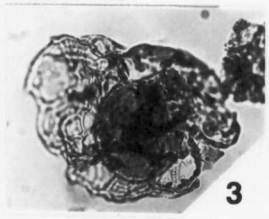
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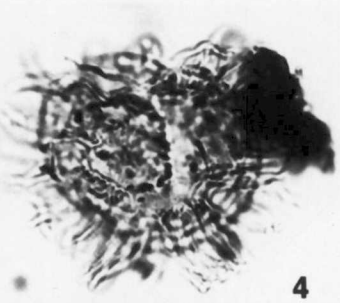
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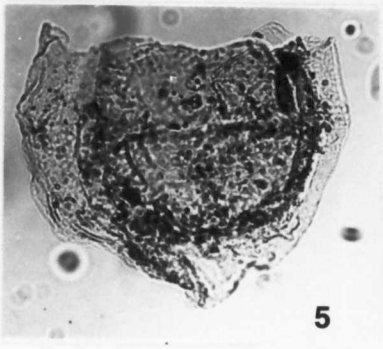
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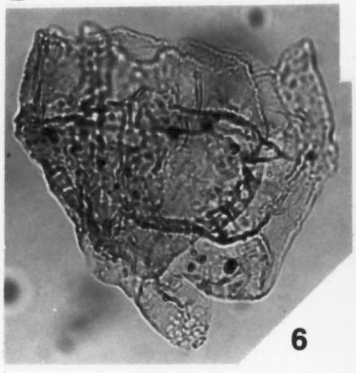
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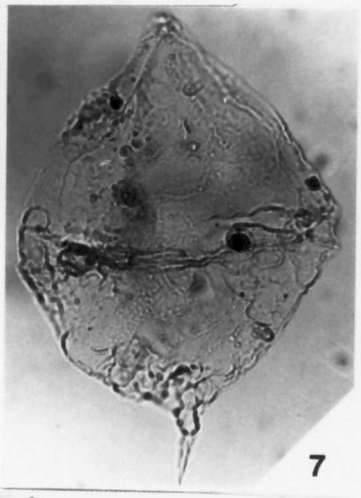
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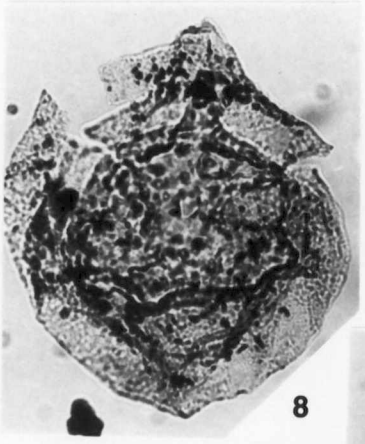
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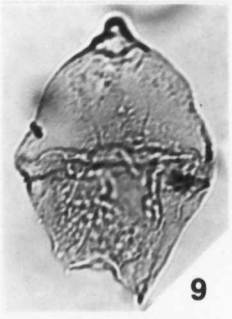
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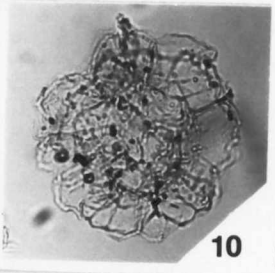
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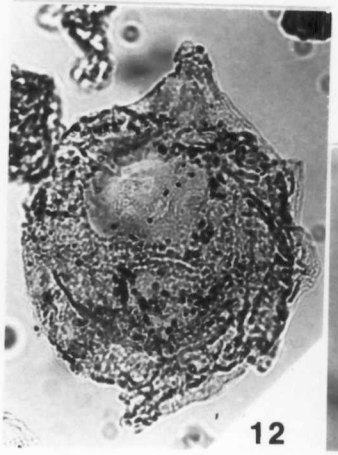
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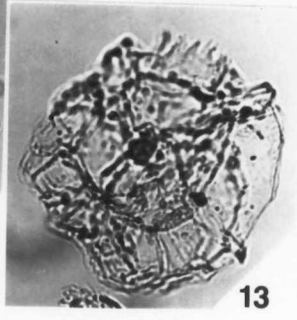
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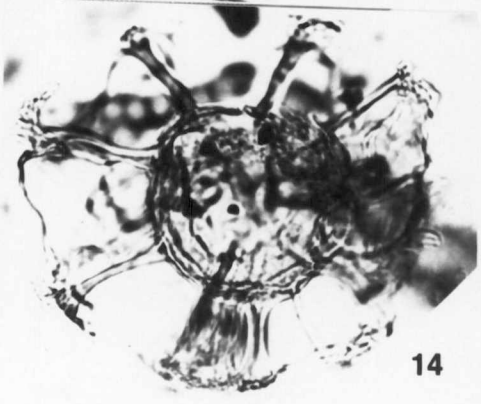
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PLATE 7

1. Unknown sp. G Slide #518, h. 14.2, v. 14.2

2. *Odontochitinopsis* sp. A Slide no. 521, h. 13.4, v. 12.2. 64 x 48 Microns.

3. *Xenascus ceratioides* Slide no. 505, h. 12.9, v. 12.7.

4. *Apteodinium cribrosum* Slide no. 503, h. 9, v. 12.3. 56 x 46 Microns.

5. *Polysphaeridium subtile* Slide no. 513, h. 29.0, v. 12.7. 90 Microns Inner Cyst.

6. Unknown sp. 516 Slide no. 516, h. 7.0, v. 13.7. 76 x 70 Microns.

7. *Xenascus ceratioides* Slide no. 594, h. 10.4, v. 6.3. 90 Microns.

8. Unknown sp. B Slide no. 491, h. 8.3, v. 19.6. 80 Microns Inner Cyst.

9. Unknkown sp. A Slide no. 501, h, 15.5, v. 6.2. 90 x 70 Microns.

10. *Xenascus ceratioides* Slide no. 498, h. 20.2, v. 20.1. 260 Microns.

11. *Exochosphaeridium phragmities* Slide no. 505, h. 13.8, v. 10.0. 80 Microns Inner Cyst.

12. *Exochosphaeridium bifidum* Slide no. 501, h. 12.6, v. 16.8. 60 Microns Inner Cyst.

13. Unknown sp. X Slide no. 509, h. 11.5, v. 10.2. 84 x 56 Microns.

PLATE 7

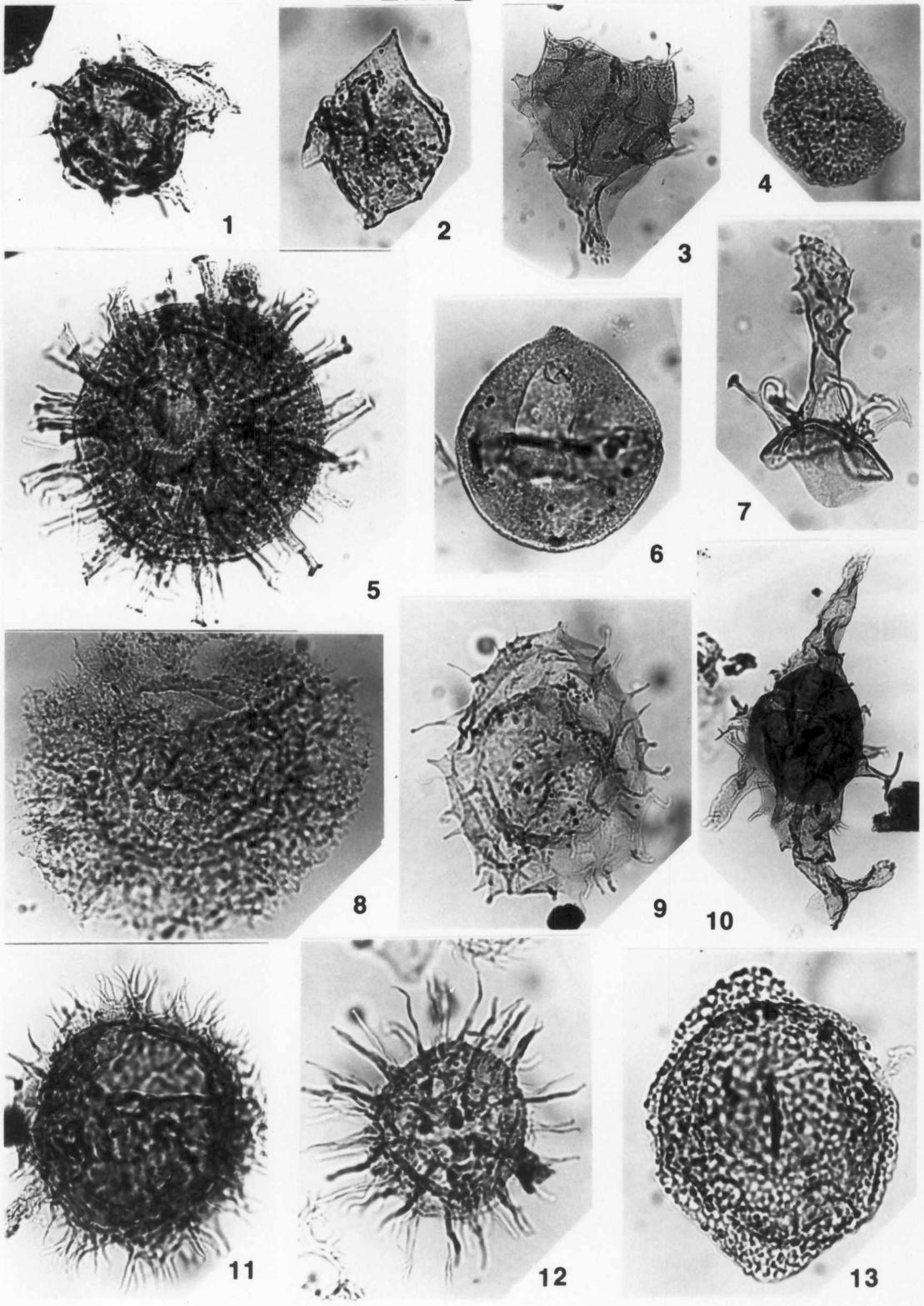


PLATE 8

1. Heterosphaeridium conjunctum Slide no. 498, h. 10.3, v. 9.6. 71 x 56 Microns Inner Cyst.

2. Florentinia sp. B Slide no. 504, h. 6.2, v. 9.9. 100 x 60 Microns.

3. Unknown Gonyaulacoid Slide no. 523, h. 26.8, v. 4. 116 x 76 Microns.

4. Heterosphaeridium conjunctum Slide no. 497, h. 14.0, v. 8.7. 60 Microns Inner Cyst.

5. Glaphyrocysta intricata Slide no. 508, h. 7.0, v. 13.0. 70 Microns Inner Cyst.

6. Cordosphaeridium fibrospinosum Slide no. 508, h. 16.7, v. 10.2. 60 Microns Inner Cyst.

7. Unknown sp. C Slide no. 501, h. 2.9, v. 16.6. 86 x 66 Microns.

8. Coronifera oceanica Slide no. 500, h. 6.5, v. 16.6. 84 x 44 Microns.

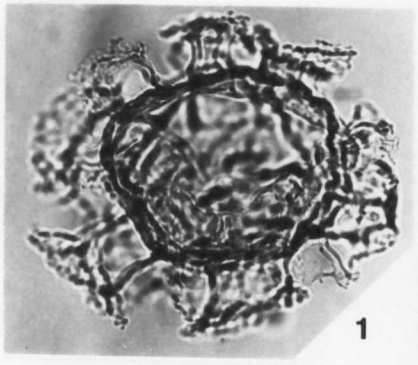
9. Surculosphaeridium longifurcatum slide no. 504, h. 7.6, v. 3.9. 58 Microns Inner Cyst.

10. Cribroperidinium edwardsii Slide no. 499, h. 16.6, v. 21.0. 128 x 88 Microns.

11. Cribroperidinium edwardsii Slide no. 499, h. 6.6, v. 3.1. 122 x 82 Microns.

12. Surculosphaeridium longifurcatum Slide no. 501, h. 22.7, v. 19.6. 60 Microns Inner Cyst.

PLATE 8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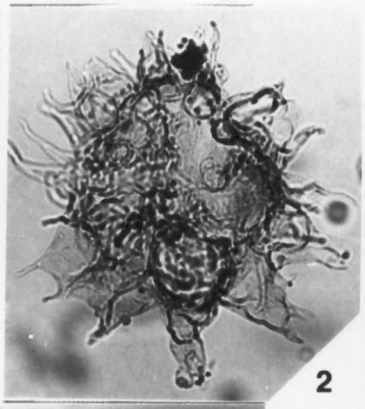
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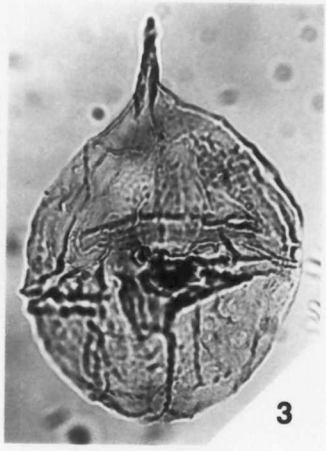
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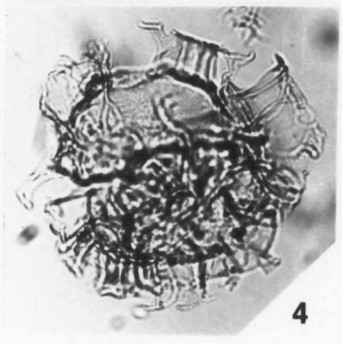
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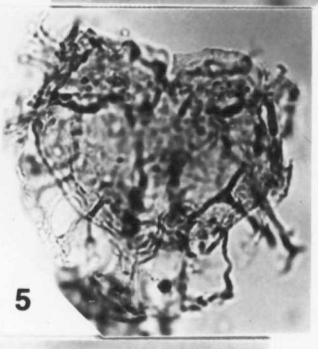
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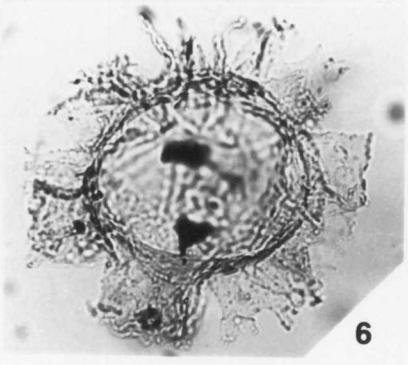
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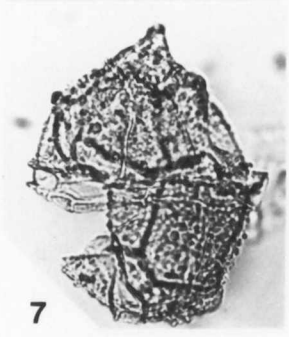
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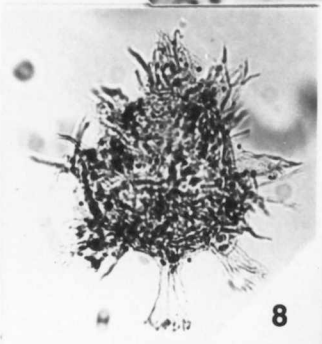
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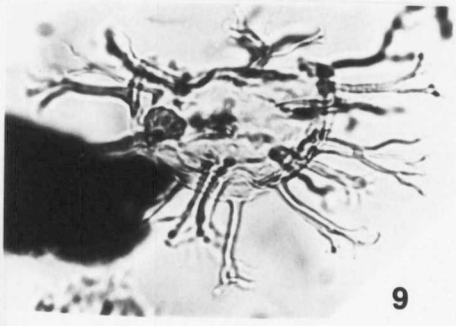
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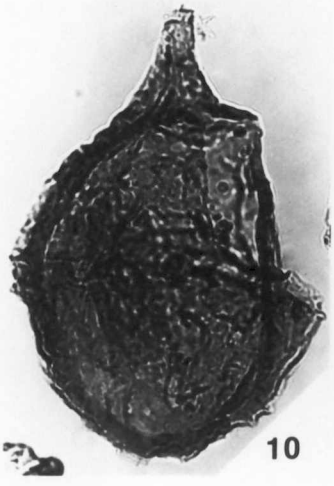
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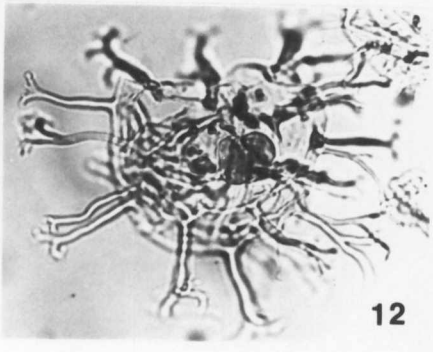
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PLATE 9

1. Dinogymnium sp. B Slide no. 506, h. 42.6, v. 15.7. 64 x 27 Microns.

2. Dinogymnium sp. C Slide no. 514, h. 19.4, v. 11.7. 46 x 15 Microns.

3. Dinogymnium denticulatum Slide no. 504, h. 20.2, v. 15.8. 46 x 30 Microns.

4. Dinogymnium cerviculum Slide no. 507, h, 8.6, v. 10.7. 38 x 20 Microns.

5. Dinogymnium digitus Slide no. 507, h. 21.0, v. 16.2. 88 x 41 Microns.

6. Dinogymnium longicornis Slide no. 508, h. 22.7, v. 3.1. 120 x 32 Microns.

7. Dinogymnium euclaense Slide no. 507, h. 21.2, v. 14.9. 34 x 42 Microns.

8. Odontochitina costa Slide no. 508, h. 7.0, v. 1.8. 156 Microns.

9. Dinogymnium acuminatum Slide no. 505, h. 18.6, v. 10.5. 82 x 54 Microns.

10. Odontochitina costata Slide no. 598, h. 20.1, v. 10.7. 200 Microns.

11. Trigonopyxidia ginella Slide no. 497. 70 Microns.

12. Odontochitina porifera Slide no. 501. 235 Microns.

13. Canningia scabrosa Slide no. 5, h. 6.2, v. 22.0. 90 x 80 Microns.

14. Canningia sp. A Slide no. 502, h. 12.6, v. 4.0. 90 x 80 Microns.

15. Odontochitina operculata Slide no. 505, h. 16.6, v. 16.0. 180 Microns.

PLATE 9


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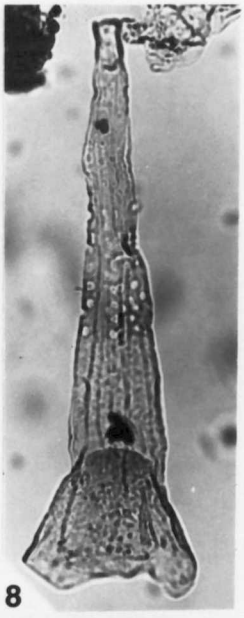
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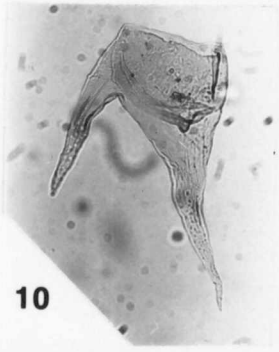
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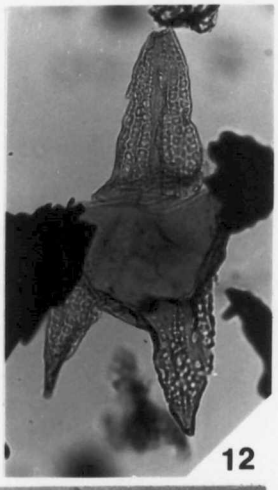
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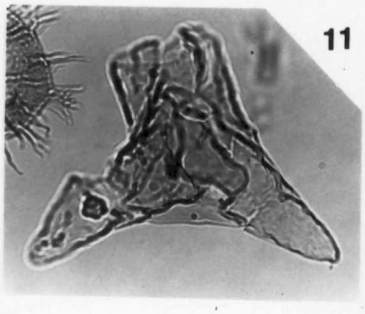
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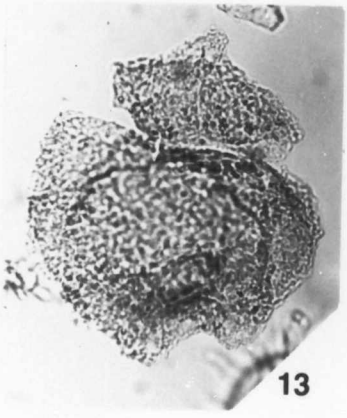
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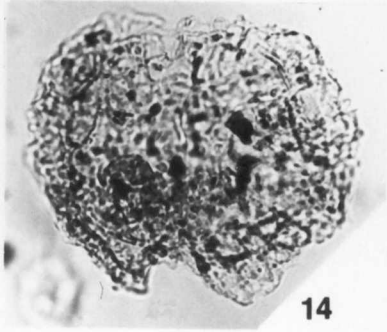
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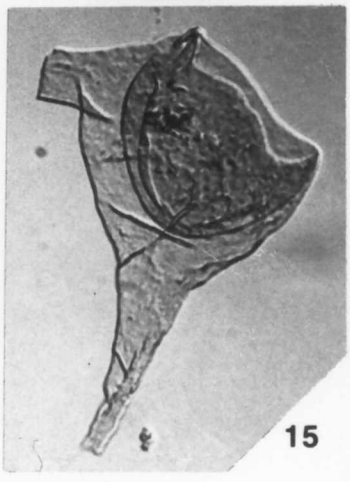
15

Appendix D

Annotated Species List

Species follow the order they are found in the Range Chart.

1. Unknown sp. A.

2. Subtilisphaera sp. *Jain and Millepied, 1973, p. 26-27; emend. Lentin and Williams,, 1976, p. 117-118.

3. Cleistosphaeridium sp. *Davey et al., 1966, p. 166.

4. Exochosphaeridium bifidum (Clarke and Verdier, 1967, p. 72, pl. 17, fig. 5-6; text-fig. 30) Clarke et al., 1968, p. 182.

5. Canningia scabrosa Cookson and eisenack, 1970a, p. 146, pl. 13, fig. 6-7.

6. Florentinia clavigera (Deflandre, 1937b, p. 71, pl. 11, fig. 1-2) Davey and Verdier, 1973, p. 192; emend. Davey and Verdier, 1976, p. 315.

7. Chatangiella victoriensis (Cookson and Manum, 1964, p. 522, pl. 76, fig. 3-8) Lentin and Williams, 1976, p. 55.

8. Chatangiella granulifera (Manum, 1963, p. 61, pl. 3, fig. 5-9) Lentin and Williams, 1976, p. 54.

9. Palaeohystrichophora infusorioides Deflandre, 1935, p. 230-231, pl. 8, fig. 4.

10. Palaeohystrichophora sp. B. *Deflandre, 1935, p. 230; emend. Deflandre and Cookson, 1955, p. 257.

11. Lejeunecysta sp. A. *Artzner and Dorhofer, 1978, p. 1381; emend. Lentin and Williams, 1976, p. 68; emend. Bujak, 1980, p. 68.

12. Spinidinium echinoideum (Cookson and Eisenack, 1960a, p. 2, pl. 1, fig. 5-6) Lentin and Williams, 1976, p. 64.

13. Spinidinium essoi Cookson and Eisenack, 1967a, p. 135, pl. 19, fig. 1-8.

14. Odontochitina proifera Cookson, 1956, p. 188, pl. 1, fig. 17.

15. Coronifera oceanica Cookson and Eisenack, 1958a p. 45, pl. 12, fig. 6; emend. May, 1980, p. 48-49.

16. Xenascus ceratioides (Deflandre, 1937b, p. 66-67, pl. 12, fig. 7-8) Lentin and Williams, 1973, p. 144.

17. Cyclonephelium distinctum Deflandre and Cookson, 1955, p. 285-286, pl. 2, fig. 14; text-fig. 47-48.

18. Polysphaeridium subtile Davey and Williams, 1966b, p. 92, pl. 11, fig. 2; emend. Bujak et al., 1980, p. 34.

19. Trigonopyxidia ginella (Cookson and Eisenack, 1960a, p. 11, pl. 3, fig. 18-20) Downie and Sarjeant, 1965, p. 149.

20. Dinogymnium acuminatum Evitt et al., 1967, p. 8-16, pl. 1&2; pl. 3, fig. 1-8, 10, 12, 20; text-fig. 11-23.

21. Spiniferities ramosus (Ehrenberg, 1838, pl. 1, fig. 1-2, 5) Loeblich and Loeblich, 1966, p. 56-57.

22. Odontochitina operculata (O. Wetzel, 1933a, p. 170, pl. 2, fig. 21-22; text-fig. 3) Deflandre and Cookson, 1955, p. 291-292.

23. Cribroperidinium edwardsii (Cookson and Eisenack, 1958, p. 32, pl. 3, fig. 5-6; text-fig. 7).

24. Phelodinium tricuspis (O. Wetzel, 1933a, p. 166, pl. 2, fig. 14) Stover and Evitt, 1978, p. 118.

25. Isabelidinium microarmum (McIntyre, 1975, p. 65, pl. 1, fig. 5-8) Lentin and Williams, 1977a, p. 168.

26. Apteodinium cribrosum Cookson and Eisenack, 1968, p. 112, fig. 1L.

27. Senoniasphaera rotundata Clarke and Verdier, 1967, p. 62-63, pl. 14, fig. 1-3; text-fig. 25.

28. Spiniferities granulatus (Davey, 1969b, p. 4-5, pl. 1, rig. 4-7) Lentin and Williams, 1973, p. 128.

29. Hystrichokolpoma rigaudae Deflandre and Cookson, 1955, p. 279-281, pl. 6, fig. 6, 10.

30. Rottnestia borussica (Eisenack, 1954, p. 62, pl. 9, fig. 5-7) Cookson and Eisenack, 1961b, p. 42.

31. Unknown sp. B.

32. Spiniferites cingulatus (O. Wetzel, 1933b) Sarjeant, 1970, p. 76.

33. Trithyrodinium sp. A. *Drugg, 1967, p. 20; emend. Lentin and Williams, 1976, p. 98.

34. Alterbia acutula (Wilson, 1967b, p. 225-226, rig. 11-12) Lentin and Williams, 1976, p. 48.

35. Unknown sp. C.

36. Florentinia sp. A. *Davey and Verdier, 1973, p. 185-186; emend. Duxbury, 1980, p. 119.

37. Florentinia laciniata Davey and Verdier, 1973, p. 186-187, pl. 2, fig. 1, 3-4, 6-7, 9.

38. Senoniasphaera protrusa Clarke and Verdier, 1967, p. 61-62, pl. 14, fig. 7-9; test-fig. 24.

39. Operculodinium sp. A. *Wall, 1967, p. 110-111.

40. Fromea sp. *Cookson and Eisenack, 1958, p. 55.

41. Exochosphaeridium phragmities Davey et al., 1966, p. 165-166, pl. 2, rig. 8-10.

42. Hystrichosphaeridium palmatum (White, 1842, p. 39, pl. 4, fig. 12 ex Bronn, 1848, p. 1375) Downie and Sarjeant, 1965, p. 121.

43. Spinidinium clavus Harland, 1973, p. 674-675, pl. 84, fig. 5-6, 10; text-fig. 9.

44. Dinogymnium euclaense Cookson and Eisenack, 1970a, p. 139, pl. 10, fig. 9-12.

45. Heterosphaeridium conjunctum Cookson and Eisenack, 1968, p. 115; text-fig. 4G-H.

46. Isabelladinium acuminatum (Cookson and Eisenack, 1958, p. 27, pl. 4, fig. 5-8) Stover and Evitt, 1978, p. 109.

47. Trithyrodinium sp. B. *Drugg, 1967, p. 20; emend. Lentin and Williams, 1976, p. 98.

48. Ceratiopsis pannucea (Stanley, 1965, p. 220, pl. 22, fig. 1-4, 8-10) Lentin and Williams, 1977b, p. 21.

49. Ceratiopsis diebelii (Alberti, 1959b, p. 99, pl. 9, fig. 18-21) Vosshennikova, 1967, p. 159.

50. Laciniadinium arcticum (Manum and Cookson, 1964, p. 18-19, pl. 6, fig. 1-4) Lentin and Williams, 1981, p. 41.

51. Surculosphaeridium longifurcatum (Firtion, 1952, p. 157, pl. 9, fig. 1; test-fig. H-M) Davey et al., 1966, p. 163.

52. Diphyes sp. A. *Cookson, 1965a, p. 85; emend. Davey and Williams, 1966b, p. 95-96.

53. Florentinia sp. B. *Davey and Verdier, 1973, p. 185-186; emend. Dorhofer and Davies, 1980, p. 36; emend. Below, 1981, p. 30.

54. Canningia sp. A. *Cookson and Eisenack, 1960b, p. 251; emend. Dorhofer and Davies, 1980, p. 36.

55. Hystrichosphaeridium tubiferum (Ehrenberg, 1838, pl. 1, fig. 16) Deflandre, 1937b, p. 68; emend. Davey and Williams, 1966b, p. 56-58.

56. Palaeocystodinium sp. C. *Alberti, 1961, p. 20.

57. Hystrichosphaeridium sp. X. *Deflandre, 1937b, p. 68; emend. Davey and Williams, 1966b, p. 55-56.

58. Areoligera senonensis Lejeune-Carpentier, 1938, p. 164-166, test-fig. 1-3. Senonian.

59. Cyclonephelium sp. D. *Deflandre and Cookson, 1955, p. 285; emend. Williams and Downie, 1966c, p. 223 . . . Emend. Stover and Evitt, 1978, p. 35.

60. Odontochitina costata Alberti, 1961, p. 31, pl. 6, fig. 10-13; emend. Clarke and Verdier, 1967, p. 58-59.

61. Hystrichosphaeridium sp. A. *Deflandre, 1937b, p. 68; emend. Davey and Williams, 1966b, p. 55-56.

62. Glaphyrocysta intricata (Eaton, 1971, p. 365, pl. 4, fig. 8-10) Stover and Evitt, 1978, p. 50.

63. Chatangiella tripartita (Cookson and Eisenack, 1960a, p. 2, pl, 1, fig. 20) Lentin and Williams, 1976, p. 55.

64. Tanyosphaeridium xanthiopyxides (O. Wetzel, 1933b, p. 44, pl. 4, fig. 25) Stover and Evitt, 1978, p. 85.

65. Nematosphaeropsis pusulosa (Morgenroth, 1966b, p. 8, pl. 2, fig. 6) Stover and Evitt, 1978, p. 176.

66. Hystrichosphaeridium tubiferum subsp. brevispinum (Davey and Williams, 1966b, p. 38, pl. 10, fig. 20) Lentin and Williams, 1973, p. 80.

67. Cyclonephelium compactum Deflandre and Cookson, 1955, p. 285, pl. 2, fig. 11-13; test-fig. 44-46.

68. Palaeoperidinium sp. A. *Deflandre, 1935, p. 227; emend. Sarjeant, 1967b, p. 246.

69. Isabelidinium belfastense (Cookson and Eisenach, 1961a, p. 71, pl. 11, fig. 4-6) Lentin and Williams, 1977a, p. 167.

70. Eatonicysta sp. A. *Stover and Evitt, 1978, p. 41.

71. Ginginodinium sp. A. *Cookson and Eisenack, 1960a, p. 7; emend. Lentin and Williams, 1976, p. 95-96.

72. Hystrichosphaeridium sp. 512. *Deflandre, 1937b, p. 68; emend Davey and Williams, 1966b, p. 58-59.

73. Impagidinium sp. A. *Stover and Evitt, 1978, p. 165.

74. Alterbia sp. A. *(Lentin and Williams, 1976, p. 47, nom, subst. pro Albertia Vozzhennikova, 1967, (non Albertia Schimper, 1827.)

75. Dinogymnium sp. A. *Evitt et al., 1967, p. 4-8.

76. Spinidinium sverdrupianum (Manum, 1963, p. 59-60, pl. 2, fig. 6-15; text-fig. 3) Lentin and Williams, 1973, p. 64.

77. Trithyrodinium sp. C. *Drugg, 1967, p. 20; emend. Lentin and Williams, 1976, p. 98.

78. Dinogymnium longicornis (Vozzhennikova, 1967, p. 46, pl. 1, fig. 8; pl. 3, rig. 6; pl. 4, fig. 6a-b, 7) Harland, 1973, pl 678.

79. Unknown X

80. Unknown Peridinioid 518.

81. Palaeocystodinium sp. A. *Alberti, 1961, p. 20.

82. Alterbia sp. B. *Lentin and Williams, 1976, p. 47 nom, subst. pro Albertia Vozzhennikova, 1967 (non Albertia Schimper, 1827).

83. Aiora sp. *Cookson and Eisenack, 1960a, p. 9.

84. Dinogymnium digitus (Deflandre, 1935, p. 225, text-fig. 7-8) Evitt et al., 1967, p. 18-19.

85. Cordosphaeridium fibrospinosum Davey and Williams, 1966b, p. 86, pl. 5, fig. 5.

86. Dinogymnium cerviculum Cookson and Eisenack, 1970a, p. 138, pl. 10, fig. 6.

87. Spinidinium sp. B. *Cookson and Eisenack, 1962b, p. 489; emend. Lentin and Williams, 1976, p. 62-63.

88. Palaeocystodinium benjaminii Drugg, 1967, p. 31, pl. 3, fig. 1; pl. 9, fig. 3.

89. Lejeunecysta sp. B. *(Gerlach, 1961, p. 169, pl. 26, fig. 10-11; emend. Kjellstrom, 1972, p. 469) Artzner and Dorhofer, 1978, p. 1381.

90. Palaeohystrichophora sp. C. *(Deflandre, 1935, p. 230; emend. Deflandre and Cookson, 1955, p. 257.

91. Adnatosphaeridium vittatum Williams and Downie, 1966c, p. 215, pl. 24, fig. 3, 7; text-fig. 56.

92. Spinidinium densispinatum Stanley, 1965, p. 226-227, pl. 21, fig. 1-5.

93. Dinogymnium sp. B. *Evitt et al., 1967, p. 4-8.

94. Unknown sp. 506.

95. Deflandre sp. C. *Eisenack, 1938, p. 187, emend. Williams and Downie, 1066c, p. 231; emend, Lentin and Williams, 1976, p. 35-36.

96. Ascodinium acrophorum Cookson and Eisenack, 1960a, p. 5, pl. 1, fig. 19-20.

97. Unknown sp. 516.

98. Dinogymnium denticulatum (Alberti, 1961 p. 5, pl. 3, fig. 2-3) Evitt et al., 1967, p. 18.

99. Oligosphaeridium sp. *Davey and Williams, 1966b, p. 70-71.

100. Dinogymnium sp. C. *Evitt et a., 1967, p. 4-8.

101. Unknown, sp. G.

102. Alterbia sp. C. *Lentin and Williams, 1976, p. 47 nom, subst. pro Albertia Vozzhennikova, 1967 (non Albertia Schimper, 1827).

103. Dinogymnium sibiricum (Vozzhennikova, 1967, p. 47-58, pl. 2, fig. 2-3b; pl. 3, fig. 2-3) Lentin and Williams, 1973, p. 50.

104. Cyclonephelium sp. C. *Deflandre and Cookson, 1955, p. 285; emend. Williams and Downie, 1966c, p. 223 . . . emend. Stover and Evitt, 1978, p. 35.

105. Odontochitinopsis sp. A. *Eisenack, 1961, p. 298-299, 308.

106. Unknown Peridinioid.

107. Isabeladinium sp. A. *Lentin and Williams, 1977a, p. 167, (nom. subst. pro Isabelia Lentin and Williams, 1976, p. 56).

108. Deflandre sp. A. *Eisenack, 1938, p. 187; emend. Williams and Downie, 1966c, p. 231; emend. Lentin and Williams, 1976, p. 35-36.

109. Deflandre sp. B. *Eisenack, 1938, p. 187; emend. Williams and Downie, 1966c, p. 231; emend. Lentin and Williams, 1976, p. 35-36.

110. Unknown sp. Z.

111. Unknown Gonyaulacoid.

112. Unknown Deflandre *(Eisenack, 1938, p. 187; emend. Williams and Downie, 1966c, p. 231; emend. Lentin and Williams, 1976, p. 35-56.

113. Spinidinium sp. G. *Cookson and Eisenack, 1962b, p. 489; emend. Lentin and Williams, 1976, p. 62-63.

114. Unknown sp. E.

Appendix E

Dinoflagellate Cyst Arranged by Family, Genus, and Species

CLASS DINOPHYCEAE
ORDER GYMNODINIACEAE
Family Gymnodiniaceae

Dinogymnium acuminatum
Dinogymnium cerviculum
Dinogymnium denticulatum
Dinogymnium digitus
Dinogymnium euclaense
Dinogymnium longicornis
Dinogymnium sibiricum
Dinogymnium sp. A.
Dinogymnium sp. B.
Dinogymnium sp. C.

ORDER PERIDINIALES
Family Gonyaulacaceae

Adnatosphaeridium vittatum
Aiora sp.
Apteodinium cribrosum
Areoligera senonensis
Cleistophaeridium sp.
Cordosphaeridium fibrospinosum
Coronifera oceanica
Cribroperdinium edwardsii
Cyclonephelium compactum
Cyclonephelium distinctum
Cyclonephelium sp. C.
Cyclonephelium sp. D.
Diphyes sp. A.
Exochosphaeridium bifidum
Exochosphaeridium phragmitties
Eatonicysta sp. A.
Florentinia clavigera
Florentina laciniata
Florenitina sp. A.
Florentina sp. B.
Glaphyrocysta intricata
Heterosphaeridium conjunctum
Hystrichosphaeridium palmatum
Hystrichokolpoma rigaudae
Hystrichosphaeridium tubiferum
Hystrichosphaeridium tubiferum subsp. brevispinum
Hystrichosphaeridium sp. A.
Hystrichosphaeridium sp. X.

Hystrichosphaeridium sp. 512.
Impagidinium sp. A.
Nematosphaeropsis pusulosa
Oligosphaeridium sp.
Operculodinium sp. A.
Polysphaeridium subtile
Rottinestia borussica
Senoniasphaera protrusa
Senoniasphaera rotundata
Spiniferites cingulatus
Spiniferities granulatus
Spiniferities ramosus
Surculosphaeridium longifurcatum
Tanyosphaeridium xanthiopyxides
Unknown gonyaulacoid
Unknown sp. A.
Unknown sp. C.
Unknown sp. Z.

Family Peridiniaceae

Alterbia acutula
Alterbia sp. A.
Alterbia sp. B.
Alterbia sp. C.
Ascodinium acrophorum
Ceratiopsis pannucea
Ceratiopsis diebelii
Chatangiella granulifera
Chatangiella tripartita
Chatangiella victoriensis
Deflandre sp. A.
Deflandre sp. B.
Deflandre sp. C.
Ginginodiniun sp. A.
Isabelladinium acuminatum
Isabelidinium belfastense
Isabelidinium microarmum
Isabeladinium sp. A.
Laciniadinium arcticum
Lejeunecysta sp. A.
Lejeunecysta sp. B.
Palaeohystrichophora infusorioides
Palaeohystrichophora sp. B.
Palaeohystrichophora sp. C.
Palaeocystodinium benjamimii
Palaeocystodinium sp. A.
Palaeocystodinium sp. C.

Palaeoperidinium sp. A.
Phelodinium tricuspis
Spinidinium clavus
Spinidinium densispinatum
Spinidinium echinoideum
Spinidinium essoi
Spinidinium sverdrupianum
Spinidintum sp. B.
Spinidinium sp. G.
Subtilisphaera sp.
Trithyrodinium sp. A.
Trithyrodinium sp. B.
Trithyrodinium sp. C.
Unknown deflandre
Unknown peridinioid 518
Unknown sp. 516
Unknown sp. 506
Unknown peridinioid

Unknown Family Association

Canningia scabrosa
Canningia sp. A.
Fromea sp.
Odontochitina costata
Odontochitina operculata
Odontochitina porifera
Odontochitinopsis sp. A.
Trigonopyxidia ginella
Xenascus ceratioides
Unknown sp. B.
Unknown sp. E.
Unknown sp. G.
Unknown X

VITA

Christian (Chris) J. Heine was born in Pittsburgh, Pennsylvania on October 30, 1956. He attended two local elementary schools and junior high before entering Bethel Park Senior High, graduating in the spring of 1974. He entered Pennsylvania State University where he received a B.S. in Forestry in June 1978. After graduation, he became an employee of Koppers Company, Inc. and was assigned to work as a Forester in East Tennessee. In the fall of 1982, he entered The University of Tennessee where, after completing undergraduate course requirements in the geology major, he was awarded a graduate assistantship and began graduate studies. He received the Masters of Science degree in June of 1986.

After graduation, Mr. Heine accepted a position with Mobil Exploration and Producing Services Inc., in Dallas, Texas where he had previously served two summer internships during the period of his graduate studies.