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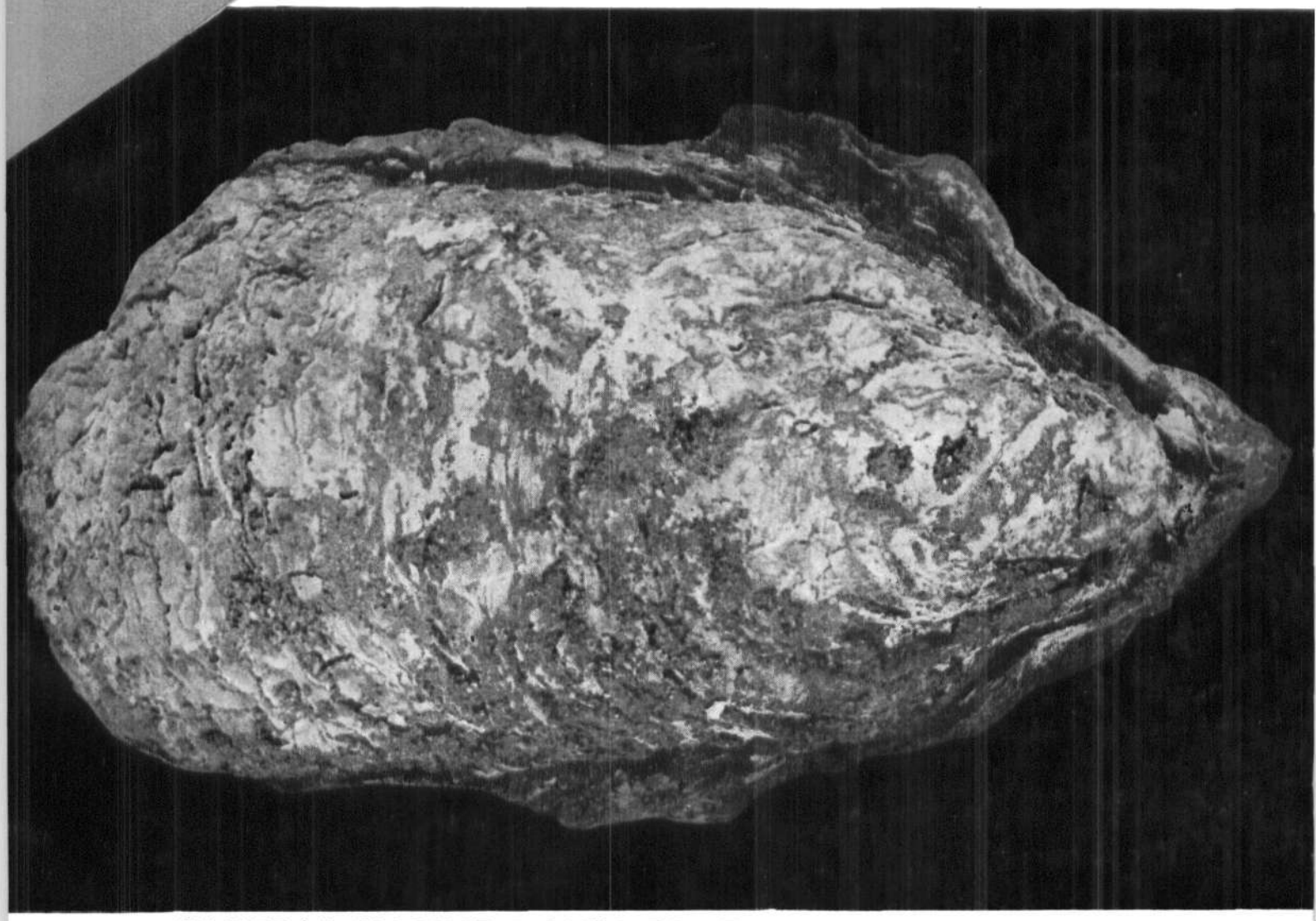
Edited by : K. NAGAPPAN NAYAR AND S. MAHADEVAN



CENTRAL MARINE FISHERIES RESEARCH INSTITUTE
(Indian Council of Agricultural Research)
P.B. No. 2704, Cochin 682 031, India

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P. S. B. R. JAMES

Director

Central Marine Fisheries Research Institute
Cochin-682 031, India

Edited by

K. NAGAPPAN NAYAR AND S. MAHADEVAN

Senior Scientists

Research Centre of Central Marine Fisheries Research Institute
Tuticorin-682 001, India

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PREFACE

Most scientific studies on oyster resources, biology and farming have been carried out in the twentieth century. New approaches to our understanding of the intricate pattern of the oyster behaviour in respect of feeding, reproduction, larval development and disease control have enabled scientists to plan and undertake highly successful oyster farming in recent years. This is not only the situation in advanced countries but has become a distinct possibility in India. Scientists of the Central Marine Fisheries Research Institute have achieved remarkable success by carrying out extensive investigations relating to the breeding and farming of the edible oyster during the past one decade. In addition to the evaluation of the potentialities of natural resources in several zones of Indian coastal waters, estuaries and backwaters, suitable technologies for oyster farming and hatchery production of oyster seed have been developed by the CMFRI. It would only be a matter of time before commercial farming of the edible oyster takes place in the country.

It is appropriate that on this occasion of the *National Seminar on Shellfish Resources and Farming* being held at Tuticorin during 19—21 January, 1987, a Bulletin on **Oyster Culture : Status and Prospects** is brought out to make available information on the present status of oyster research in India and focus attention of scientists on various problems connected with oyster resources, biology and farming. The Bulletin with 13 Chapters gives the results of more than a decade of research. The Tuticorin Research Centre of CMFRI has played the leading role in our R & D activities on oyster culture. Some work has also been done from the Mandapam Regional Centre and Kakinada and Karwar Research Centres. While the current emphasis is to diversify our research activities in terms of species and regions, the present Bulletin concentrates on the different themes worked out at the Tuticorin Research Centre.

Chapter 1 gives an account of the *taxonomy* of Indian oysters. Chapter 2 deals with the *ecology* of oyster beds. Chapters 3 and 4 identify the natural oyster *resources* along the Indian coastline. Chapters 5 and 6 relate to the investigations on the *biology* of the backwater oyster *Crassostrea madrasensis*. The rest of the chapters deal with aspects of oyster farming and seed production. Chapter 7 gives the techniques of *spat collection* from the natural beds. Chapter 8 deals with *hatchery* production of seed, an aspect of considerable importance to development of oyster culture in the country. Chapter 9 relates to *oyster farming techniques* developed at CMFRI. Chapter 10 deals with *post-harvest technology* of oyster production. Chapter 11 projects the *economics* of oyster culture. Chapter 12 gives an account of the problems of *pests and predators* in the oyster farm. Lastly, Chapter 13 gives an *overview* of oyster culture indicating the R & D thrusts required in future if oyster farming is to become a commercial reality in India. Thus a wide range of subjects on oyster resources and farming has been dealt with in the Bulletin. I have no doubt that this publication will be well received by the scientists and technicians who are engaged in oyster research in the country as well as abroad, as also by the development and extension officers who would be involved in promoting and aiding commercial oyster culture operations in future. Specifically, the prospective entrepreneurs who have several points to be considered will find the Bulletin very useful.

Concerted efforts have been made by Shri K. Nagappan Nayar and Shri S. Mahadevan, Senior Scientists of the Molluscan Fisheries Division of the Institute who have organized research on oyster culture in the Institute. In addition to writing some Chapters, they contributed greatly to editing of the bulletin. I have

great pleasure to commend their work on this occasion. Dr. K. Alagarswami, Head of Molluscan Fisheries Division took keen interest and made many helpful suggestions for which I thank him sincerely.

Dr. K. Satyanarayana Rao and Shri P. T. Meenakshisundaram have assisted in the printing of the bulletin. I express my appreciation of the efforts put in by all the Scientists, Technical Assistants of the Institute and other members of staff who have in one way or the other contributed to the development of oyster farming on an experimental scale as reflected in the present bulletin.

P. S. B. R. JAMES
Director
Central Marine Fisheries
Research Institute
Cochin

1

TAXONOMY OF INDIAN OYSTERS

K. SATYANARAYANA RAO[1]

There has been considerable disagreement on the identity of oysters due to the large variations in shape, size, texture and colour of shell which are very much influenced by the substratum and ecological conditions. As many as hundred species of living oysters and five hundred species of extinct ones were recognized initially (Korringa, 1952). Later it has been realized that most of the species were not valid. The palaeontologist Stenzel (1971) in his treatise on the systematics of oysters recognizes eight genera of living and fossilized ones, *Ostrea, Lopha, Alectryonella, Crassostrea, Saccostrea, Striostrea, Neopycnodonte* and *Hyotissa.* Oyster biologists distinguish four genera of living species of oysters *Ostrea, Crassostrea, Pycnodonta* and *Saccostrea* and this is accepted (Yonge 1960, Galtsoff 1964, Ahmed 1975).

Ranson (1948, 1950) included living species of oysters in three genera *Pycnodonta, Ostrea* and *Gryphaea* (which is synonymous with *Crassostrea*), based on the structural features of larval shell and adult. In the genus *Pycnodonta* the larval shell has equal sized valves with five teeth on the provinculum and the adult is oviparous, rectum traverses through the ventricle and promyal chamber is present. In the genus *Ostrea* the larval shell valves are of unequal size with two teeth on each side of the provinculum and the adult is larviparous, the rectum does not pass through the ventricle and promyal chamber is absent. The genus *Gryphaea* erected by Lamarck (1801) is not valid as *Gryphaea angulata* and some other species included in it were not diagnosed. The International Commission on Zoological Nomenclature (1955) in its opinion stated that the nominal species *G. angulata* was not type species of any nominal genus and the genetic name *Crassostrea* 1897 was available for use for that species. Thus the species *angulata, virginica, gigas* and others are included under the genus *Crassostrea* which is characterized by the presence of two teeth on the right valve and three teeth on the left valve in the larval shell and in the adult the shape of shell is irregular, the shell is generally attached to the substratum, the adult is oviparous, rectum does not pass through ventricle and promyal chamber is present. In the genus *Saccostrea* the umbonal cavity of the adult is deep and there are tubercles along the inner margin of the left shell valve.

TAXONOMY OF INDIAN OYSTERS

The taxonomy of Indian oysters has been studied by Hornell (1910, 1922) Annandale and Kemp (1916), Preston (1916), Gravely (1941), Satyamurthi (1956), Durve (1968), Rao (1974) and Anonymous (1984). The Indian oysters were originally referred to the genus *Ostrea* (Awati and Rai, 1931) but later included under the genus *Crassostrea* (Rao 1956, 1958, Durve 1968), as the genus *Ostrea* comprises of larviparous oysters with the characteristics mentioned earlier. Awati and Rai (1931) have identified eight species of oysters including *Ostrea* (=*Saccostrea*) *cucullata, O* (=*Crassostrea*) *gryphoides* and *O.* (*C*) *madrasensis* but the identity of some of the oyster species has to be confirmed.

In this work oyster collections obtained from different places, Visakhapatnam, Kakinada, Madras, Athankarai, Mandapam, Tuticorin, Cochin, Mulki, Coondapur, Karwar, Ratnagiri, Bombay and Sikka (Gujarat) were examined and identified. Based on the structural features, six species of oysters including five of the genus *Crassostrea, C. madrasensis, C. gryphoides, C. rivularis, C. cristagalli* and *C. folium* and one of the genus *Saccostrea, S. cucullata* were identified. The diagnostic features of the two genera and six species are dealt with here together with their affinities and distribution.

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001.

Genus *Crassostrea* Sacco

The shell valves are variable in shape and are usually elongated. The left valve situated on the lower side, is more or less cup-like and attached to the substratum while the right one is flat and functions like a cover for the former. The hinge does not have teeth and the ligament is partly external. The adductor scar is situated dorsolaterally. The gill ostia are small and rectum does not pass through ventricle. Sexes are separate but sex change takes place in some individuals and hermaphrodite oysters occur. Members of the genus are oviparous and gametes are discharged into water where fertilization takes place. Eggs are small in size. The species of the genus are euryhaline and thrive well in turbid brackish waters.

Crassostrea madrasensis (Preston)

Synonyms

Ostrea cucullata Hornell, 1910, *Madras Fish. Bull.*, **4**, 25-31.

Ostrea virginica Annandale and Kemp, 1916, *Mem. Ind. Mus.*, **5**, 329-366.

Ostrea virginiana Hornell, 1922, *Madras Fish Bull.*, **14**, 97-215.

Ostrea virginiana var. *madrasensis* Moses, 1928, *J. Bombay nat. Hist. Soc.*, **32**, 548-552.

Ostrea arakanensis Winckworth, 1931, *Proc. Malacol. Soc. London*, **19**, 188-189

Ostrea madrasensis Preston, 1916, *Rec. Indian Mus.*, **12**, 27-139.

Ostrea madrasensis Awati and Rai, 1931, *Ostrea cucullata* (The Bombay oyster). *Indian Zool. Memoir*, **III**, 107 pp.

Ostrea madrasensis Gravely, 1941, *Bull. Madras Govt. Mus.*, (*New Ser.*) *Nat. Hist. Sect.* **5** (1) : 1-112.

Ostrea madrasensis Paul, 1942, *Proc. Indian Acad. Sci.*, **15B**, 1-42.

Ostrea (*Crassostrea*) *madrasensis* Rao, 1956, *Ibid.*, **44B**, 332-356.

Ostrea madrasensis Satyamurthi, 1956, *Bull. Madras Govt. Mus.* (*New Ser.*) *Nat. Hist. Sect.*, **1** (2) Part 7, 68.

Crassostrea madrasensis Rao, 1958, Molluscan Fisheries. In *Fisheries of West Coast of India*, S. Jones, (Ed.) 55-59.

Crassostrea madrasensis Rao, 1974, Chapt. II. *In* The Commercial Molluscs of India, *Bull. Cent. Mar. Fish. Res. Inst.*, **25**, 14.

Description

The shell valves are very irregular in shape ; they are usually elongated. When spat set on flat surfaces and there is no crowding a flat shape is attained by the oysters. Those growing on uneven areas have shape of the niche where they are present and overcrowding leads to oysters with very much twisted shells.

Outer surface of shell valves has numerous foliaceous laminae with sharp edges. Width of shell 0.38 to 0.64 and thickness 0.14 to 0.36 in length. The left valve is deep and the right one slightly concave. Hinge is narrow and elongated ; it is sometimes elevated and has a medial depression in some oysters. Adductor muscle is situated subcentrally, reniform and dark purple in colour. The colour of the outer surface is grey, green or light purple depending on the area in which the oysters occur due to the presence of detritus, algae etc. The inner surface of valves is smooth, glossy and white in colour with purplish black colouration along the margins of the valves (Pl. I A, B and C).

Remarks

Ahmed (1971, 1975) considers that this species occurring in India and Pakistan is a synonym of the American oyster *Crassostrea virginica* Gmelin. The shell of *C. madrasensis* resembles *C. virginica* in shape, presence of foliaceous laminae and reniform adductor scar. The shape, sculpture and pigmentation of inner side of shell and along the edges of the mantle and tentacles of *C. virginica* are known to vary very much (Galtsoff, 1964). The shell of *C. madrasensis* is heavier than that of *C. virginica* and there is dark purplish pigmentation along the inner margin of both valves in the former species. Until further malacalogical, karyological and physiological studies are made it is desirable to recognize *C. madrasensis* as a separate species.

This species grows to a size of 212 mm, the larger ones occurring in estuarine systems. It is a typical euryhaline species and flourishes especially well in turbid brackish waters like estuaries, creeks, bays and backwaters growing to a large size with heavy meat. It is also found in sheltered areas like ports and harbours where it occurs in large numbers attached to pillars, walls of wharves and buoys and along open coasts where hard substrata like rocks or stones are present for settlement. It is found from midlittoral zone to a depth of 15-16 metres.

The species is widely distributed in India and occurs along the east coast in Bahuda estuary near Sonapur, Vishakhapatnam, Sarada estuary, Kakinada, deltas of Godavari and Krishna Rivers, Gokulapalli, Pulicat

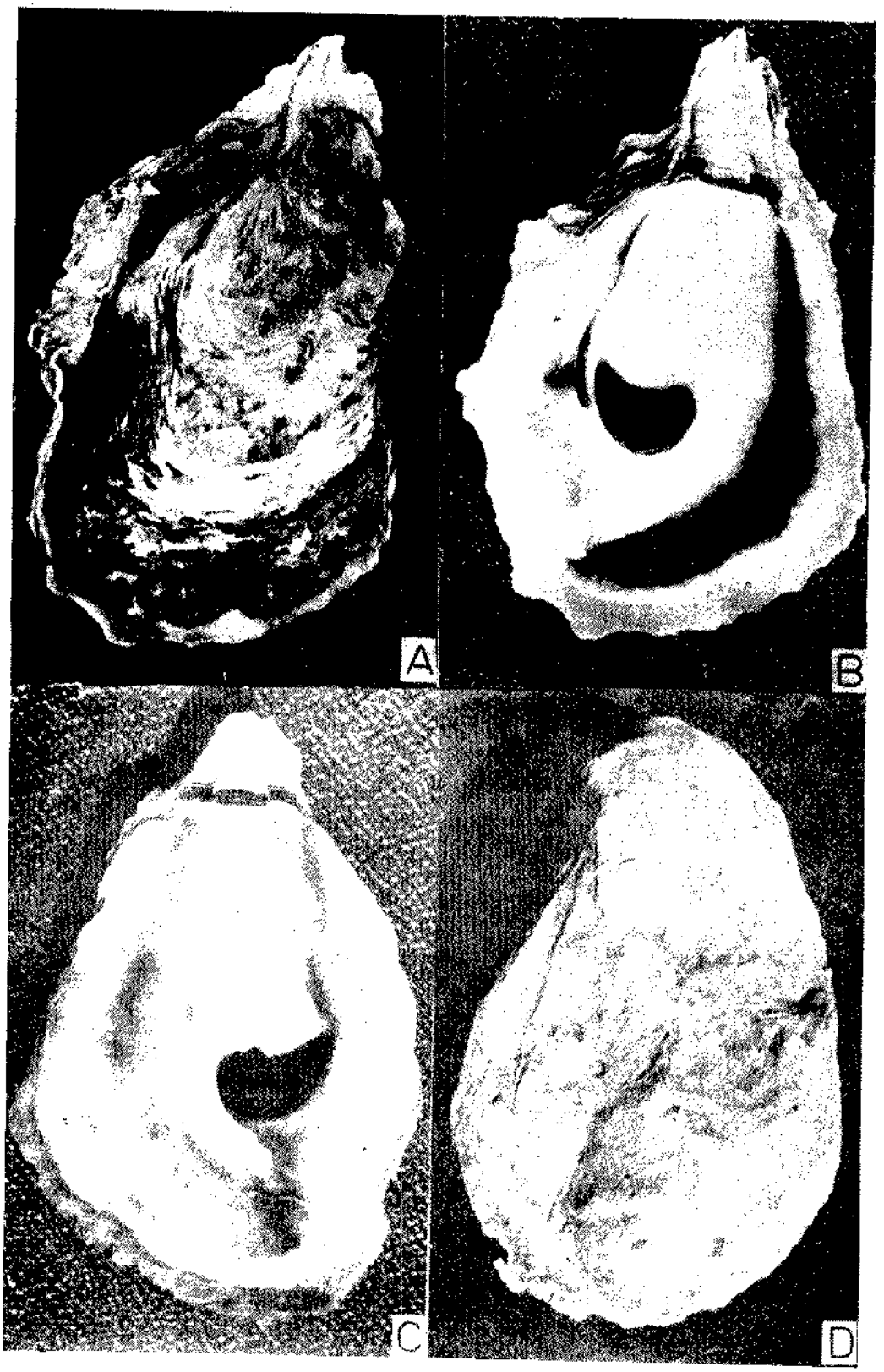

Plate I. A. *Crassostrea madrasensis* (Preston). B. Inner view of left valve of *C. madrasensis*. C. Inner view of right valve of *C. madrasensis*. D. *Crassostrea gryphoides* (Schlotheim).

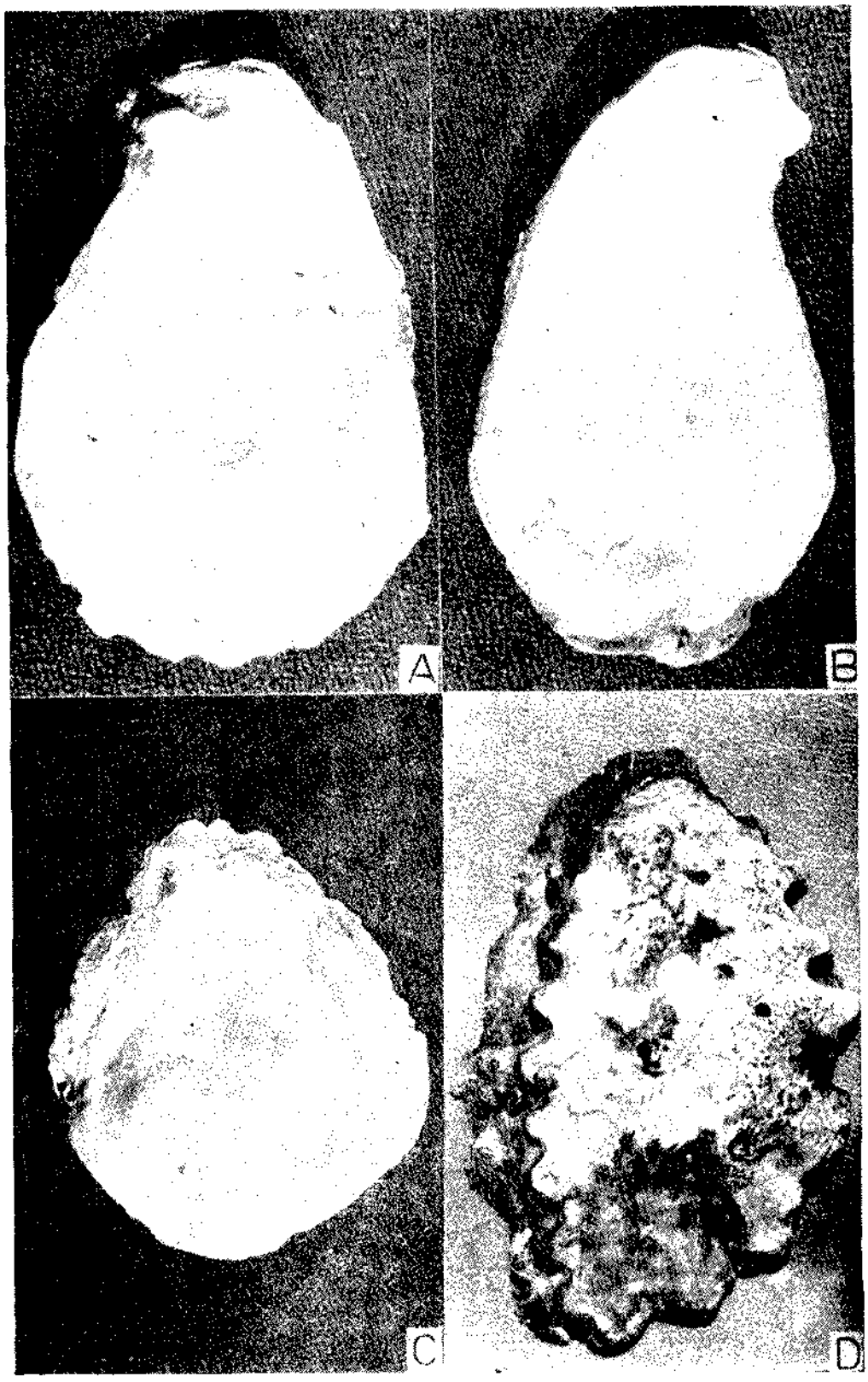

Plate II. A. Inner view of left valve of *C. gryphoides*. B. Inner view of right valve of *C. gryphoides*. C. Inner view of left valve of *Crassostrea rivularis* (Gould). D. *Saccostrea cucullata* (Born).

Lake, Ennur, Madras, Killai backwaters, Cuddalore, Muthupet, Sundarapandiapattinam, Karangad, Athankarai, Kancharangudi, Tuticorin, Pinnakayal and Palayakayal and on the west coast in Anchengo backwater, Ashtamudi and Vembanad Lakes, Cochin Harbour and backwaters, Azhikode, Beypore, Tellicherry, Elathur, Chaliyar Estuary, Pavanji, Sambhavi, Sitanadhi, Coondapur, Venkatapur, Sharavathi and Kalinadhi Estuaries and at Sikka and in Pirotan Island in Gujarat. This species occurs along Pakistan coast also (Ahmed 1971, 1975).

Crassostrea gryphoides (Schlotheim)

Synonyms

Ostrea gryphoides Vredenburg, 1904, *Rec. Geol. Surv. India*, **31**, 174-176.

Ostrea gryphoides Awati and Rai, 1931, *Ostrea cucullata* (The Bombay oyster), *Indian Zool. Memoir*, **III**, 6-7.

Crassostrea gryphoides Durve and Bal, 1961, *J. Zool, Soc. India*, **13**, 70.

Crassostrea gryphoides Rao 1974, Ch. II, In *The Commercial Molluscs of India, Bull. Cent. Mar. Fish. Res. Inst.*, **25**, 27-28.

Description

The shell valves are elongate and thick. Width of shell 0.60 to 0.72 and thickness 0.30-0.31 in length. Left valve cup like, hinge area is well developed and has a deep median groove with lateral elevations. Denticles are not present on the inner margins of valves. Adductor muscle scar is broad, more or less oblong and striations on the scar are obscure or absent. The inner surface of valves and adductor scar are pearly white in colour (Pl. I D, Pl. II A and B).

Remarks

The species grows to a size of 170 mm and occurs in the intertidal zone and down upto a depth of seven metres. Like *C. madrasensis* it is a euryhaline species, and inhabits coastal waters, estuaries and creeks of Goa, Maharashtra and Gujarat. Beds of the species are found in several places in Maharashtra such as Malad, Boiser and Satpuri Creeks, Palghar, Mahim, Kelwa, Navapur, Utsali, Dahisar, Alibag, Ratnagiri, Jaytapur and Malwan (Alagarswami and Narasimham, 1973). The species occurs at several places along the Bombay coast but does not grow more than about 40 mm in length due to heavy pollution of littoral waters. It is found at Dwarka, Aramda, Sikka, Poshetra, Balapur, Nora Island, Baida Island and Azad Island in Gujarat (Sarvaiya and Chhaya, 1983).

Crassostrea rivularis (Gould)

Synonyms

Ostrea discoidea Awati and Rai, 1931, *Ostrea cucullata* (The Bombay Oyster), *Indian. Zool. Memoir*, **III**, 7.

Ostrea rivularis Cahn, 1950, In *Oyster Culture in Japan*, **12**.

Crassostrea discoidea Rao, 1958, In *Fisheries of West Coast of India*, S. Jones (Ed.) 55-59.

Crassostrea discoidea Alagarswami and Narasimham, 1973, *Proc. Symp. Living Resources around India*, 654.

Crassostrea discoidea Rao, 1974. Ch. II. In *The Commercial Molluscs of India, Bull. Cent. Mar. Fish. Res. Inst.*, **25**, 36.

Crassostrea rivularis Imai, 1977, In *Aquaculture in Shallow Seas: Progress in Shallow Sea Culture* 125-126.

Description

This species is characterized by large, roughly round, flat, thick shell valves with a shallow shell cavity. Width of shell 0.77 to 0.89 and thickness 0.27-0.29 in length. The left valve is thick and slightly concave and the right one is about the same size or slightly larger. Adductor muscle scar is oblong and white or smoky white in colour. Inner surface of valves is white and bright (Pl. II C).

Remarks

This species can be distinguished by the shape of the shell valves, shallow shell cavity and oblong white adductor scar. The species is synonymous with *O.* (=*C*). *discoidea* recorded by Awati and Rai (1931) along Bombay coast as the structural features are similar. Imai (1977) has stated that the hinge part of the shell of *C. rivularis* is violet brown in colour. The colouration may be caused by ecological conditions such as luxuriant growth of sea weeds in the vicinity or some other such factor and therefore should not be considered a diagnostic character of taxonomic importance. The species grows to a size of 150 mm and inhabits the intertidal zone of coastal waters and creeks. It occurs in the creeks of Kutch, Aramda Creek, Poshetra Point, Port Okha, Dwarka and Porbundar in Gujarat and at Mahim, Ratnagiri and Jaytapur in Maharshtra. Outside India it is distributed along Pakistan coast (Ahmed, 1975), China and Japan (Cahn 1950, Imai 1977).

Crassostrea cristagalli (Linnaeus)

Synonyms

Mytilus cristagalli Linnaeus, 1758, *Systema Naturae*, 10th Ed.

Ostrea cristagalli Lamarck, 1819, *Histoire Naturelle des Animaux Saus Vertebres* 6 (1).

Ostrea (Lopha) cristagalli Standen and Leicester, 1906, *Report to the Govt. of Ceylon on the Pearl Oyster Fisheries of the Gulf of Mannar*, W. A. Herdman, Ed., V, 267-294.

Ostrea crista-galli Hornell, 1922, *Madras Fish. Bull.*, **14**, 97-215.

Ostrea cristagalli Prashad, 1932, *Monogr. Siboga Exped.* **29**, 1-353.

Ostrea cristagalli Satyamurthy, 1956, *Bull. Madras Govt. Mus. (New Ser.) Nat. Hist. Sect.* **1** (2) Part 7, 68-69.

Crassostrea crista-galli Rao 1974, Ch. II. *In* The Commercial Molluscs of India, *Bull. Cent. Mar. Fish. Fish. Res. Inst.*, **25** : 36-37.

Description

The shell is broadly rounded or subquadrate in shape with the margins of valves thrown into deep, sharp, angular folds and interlocking with each other. The outer surface of the folded margins has diverging, closely set, granulated striae. The external shell colour varies from brownish to violet and the internal surface of the valves is greyish white.

Remarks

The species known as Cock's comb oyster due to the shape of the shell can be identified by the deeply angular folds of the shell margin and the brown or purplish colouration. It occurs attached to stones or dead corals and grows to a size of 60 mm. At Poshitra Point, Okha District, Gujarat, beds of the species are found and the oysters are collected and consumed by fisherfolk locally. Stray individuals of the species occur along the Thanjavur coast, Palk Bay and Gulf of Mannar.

Crassostrea folium (Gmelin)

Synonyms

Ostrea folium Chemnitz, 1781, *Conch. Cab.*, V, 8 pl. vii, Figs. 662 and 666.

Ostrea folium Gmelin 1791, *Syst. Nat.*, Ed. XIII, 33-34.

Ostrea folium Reeve, 1873. *Conch. Icon., XVIII, Ostrea*, pl. xviii, Fig. 40.

Ostrea folium Satyamurthi, 1956, *Bull. Madras Govt. Mus.* (*New. Ser.*) *Nat. Hist. Sec.*, **1** (2) Part 7, 69.

Crassostrea folium Rao, 1974, Ch. II. *In* The Commercial Molluscs of India, *Bull. Cent. Mar. Fish. Res. Inst.*, **25** : 37.

Description

Shell broadly ovate in shape. The left valve is deeply concavely excavated along the middle to fit surface of attachment. Right valve is raised into a broad tube-like longitudinal rib in the middle corresponding to the excavation of left valve. Shell has a number of rounded folds diverging away from the rib-like elevation in the middle. The surface of valves is more or less smooth except for a few thin overlapping laminae towards the margin. Colour of valves pale grey or brownish purple externally and greyish internally.

Remarks

This species is found as solitary individuals attached to floating twigs in Pamban on the Southeast coast and in Gulf of Kutch and is not of economic value. It grows to a size of about 50 mm.

GENUS *Saccostrea* Dollfus AND Deutzenberg

Shell elongate and trigonal in shape. Shell cavity moderately deep. Umbonal cavity prominent. Tubercles present along the inner margin of the right valve with corresponding depressions in the left valve.

Saccostrea cucullata (Born)

Synonyms

Ostrea forskali Chemnitz, 1785, *Neues Systematisches Conchylien Cabinet*, 8.

Ostrea cucullata Born, 1780, *Testaeca Mus. Caes. Vindobon*, 114, pl. vi.

Ostrea cucullata Awati and Rai, 1931, *Ostrea cucullata* (The Bombay Oyster), *Indian Zool. Memoir*, III, 107 pp.

Ostrea forskalii Gravely, 1941, *Bull. Madras Govt. Mus. New. Ser. Nat. Hist. Sect.*, 5 (1) : 41.

Crassostrea cucullata, Rao, 1969, *Indian Frmng.*, **29** (9) : 41.

Crassostrea cucullata Rao, 1974, Chapt. II. *In* The Commercial Molluscs of India, *Bull. Cent. Mar. Fish. Res. Inst.*, **25** : 33.

Description

The shell valves of this species are hard and stony, and the shape is trigonal or pear shaped. The left valve is deep or moderately so. The right valve is flat or slightly convex and covers the left one like a lid. The hinge is straight, of moderate size and devoid of teeth and the umbonal cavity well developed. The margins of both valves have well developed angular folds sculptured with laminae. Small turbercles present along the inner margin of the right valve and there are corresponding pits in left valve. Adductor scar is kidney shaped, striated and white or greyish in colour. Colour of the outer surface of valves is variable being pale white, grey, light or dark brown, green or purplish. Inner surface of valves is white (Pl. II D).

Remarks

This species known as rock oyster since it is usually found attached to rocks in the intertidal zone is included in the genus *Saccostrea* due to the genetic character, the presence of tubercles along the inner margin of the right shell valve. The diagnostic features of the species are the trigonal or pear shape, the marginal angular folds of the shell and oblong adductor scar. According to Stenzel (1971), the Sydney rock oyster *Crassostrea commercialis* is a subspecies of *Saccostrea cucullata*. Stenzel considers *Saccostrea cucullata* as a complex superspecies from which different stocks have evolved. This view cannot be accepted unless there are evidences of genetic differences and reproductive isolation.

In contrast to *Crassostrea madrasensis*, *Saccostrea cucullata* is distributed in the marine environment in shallow coastal waters and creeks. The species enjoys a wide distribution and is found in East Africa, India Pakistan, northwestern Australia and Philippines (Ahmed, 1975). In India it occurs at various places along both the east and west coasts and around Andaman and Lakshadweep Islands. On the mainland it is found in Visakhapatnam, Madras, Killai back-waters, Mandapam, Krusadai Island, Pudumadam and Tuticorin on the east coast and Cochin, Pavanji Estuary, Coondapur Estuary, Kalinadhi, Goa, Costal waters and Creeks of Maharashtra and Gujarat on the west coast. Oysters of this specis are found growing attached to rocks in some creeks of Bombay coast (Sundaram, personal communication) and at Aramda, Dwarka, Adatra and Hanuman-dandi in Gujarat (Sarvaiya and Chhya, 1983).

Three species of oysters *Ostrea cornucopia*, Chemnitz, *O. glomerata* Gould and *O. belcheri* Sowerby have been reported by Awati and Rai (1931) who also refer to the record of three more species *O. crenulifera* Sowerby, *O. bicolor* Hanley and *O. lacerata* Hanley by Standen and Leicester in the Proceeding of Manchester Library and Philosophical Society, Vol. 7, 4th Series. Due to the absence of material, the species mentioned by Awati and Rai (1931) are not dealt with here.

Oysters are distributed at several places along our coasts and we have knowledge of the common species found in a number of areas where studies have been carried out. But our knowledge of the identity of the oyster species of India is far from complete. Very little information is available on the oyster populations in the northeast and northwest coasts of India. Recent work indicates that consideration of shell characteristics alone will not be helpful in correctly distinguishing species of oysters as they are highly influenced by ecological conditions and study of morphometric characters, structure of chromosomes, protein bands and chemical composition may be useful in identifying oyster species.

From a study of oysters collected from some parts of Indian coasts carried out employing above techniques Anonymous (1984) has indicated that there are five species, *Crassostrea madrasensis*, *C. gryphoides*, *C. rivularis*, *C.* (=*S.*) *cucullata* and *C.* (*S*) *crenulifera* and the first three species showed similarities in biochemical characteristics and the other two formed a different group. It has also been observed that *C. gryphoides* and *C. rivularis* are closer to one another in biochemical make up compared to *C. madrasensis*. A thorough study of oysters collected from various parts of the Indian coasts using such techniques is needed for removing confusion which exists about some species and for better understanding of the identity of the various species as well as their affinities.

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2

ECOLOGY OF OYSTER BEDS

S. MAHADEVAN[1] AND K. NAGAPPAN NAYAR[1]

INTRODUCTION

The oysters of the world are grouped into one family Ostreidae. Within this family are 3 groups or genera *viz., Ostrea, Crassostrea* and *Pycnodonta.* The genus *Ostrea* which is widespread through most part of the world is generally considered to be adapted to clear waters with little sediment and high salinity. *Crassostrea* exist in estuaries where the silt load is high and salinity variable and generally low. *Pycnodonta* is a tropical open sea form and thrives in high salinity. They are less abundant. In each of these genera are a number of species, about 100 species throughout the world The various species inhabit coastal waters within the broad belt of the seas, limited by the latitudes 22°S and 64°N. Patchy settlement cover many square miles of bottom of littoral and intertidal zones. They also thrive above the bottom attached to rocks and underwater structures, submerged branches and trunks of fallen trees, concrete embankments and piles and miscellaneous objects in the sea shore. These aggregations of live oysters and empty shells are termed as *oyster beds* or *oyster bottoms* or *oyster reefs.* Once they are densely settled they become important to man as source of food.

Oysters have some common factors in whichever country they are found. Description of oyster bottoms usually provide information about their location, nature of bottom and depth. The oyster bed is an example of 'biocoenosis' or a social community (Mobius, 1883) of living beings, a massing of individuals with ideal conditions governing their existence. This community character of the biocoenosis is affected as also the oyster by the changes in the factors of environment—natural or man made. The well being of oyster population is closely linked up with factors such as the character of bottom, water movements, salinity of water, temperture and abundance of planktonic food. Excessive sedimentation, extreme turbidity of water and pollution caused in the oyster bed areas due to techno-economic activities of man are some of the adverse factors disturbing the established biological balance. Predation, competition and out-break of enzootic and epizootic diseases also spell disaster to the oyster life and surrounding cohabiting organisms. In spite of these natural and man made disasters the oyster possesses some amount of flexibility in physiological adaptations to tolerating fairly wide range of salinity and temperature variation, feeding flexibility and prolificity of breeding and self defensive mechanisms for self perpetuation and survival.

The 'Oyster' is scientifically the best known marine animal in the world (Nelson, 1938). Baughman (1948) has provided a complete annotated bibliography of oysters and recently Joyce Jr. (1972) has updated all publications by listing 4,117 referenes. When compared to the voluminous data and knowledge available from the above and other equally important publications like that of Breisch and Kennedy (1980) brought out subsequently on the ecology of oysters it should be admitted that the attention paid in India to the study of oyster in general and ecology in particular appears very limited. This is partly due to the fact that till 1975 oyster did not figure prominently in our piroity areas of Research and Development. From the Indian point of view and interest species of *Crassostrea* are more important as the genus *Ostrea* is not reported to occur along the Indian coastline. Some investigations of importance and interest have been conducted in the early years on the Indian backwater oyster *Crassostrea madrasensis* (Preston) and *C. gryphoides.* Hornell (1910a, 1910b), Paul (1942), Rao

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001.

(1951, 1956, 1983) Rao and Nayar (1956) and Durve (1962, 1967) have given information on the oyster biology and ecology.

Proper understanding of the ecology of the oyster bed is very essential in order to evaluate the influence of different physical, chemical and biological environmental factors and their interaction. This will help in future management of oyster fishing and farming. Most of the information on the oyster ecology given presently is drawn from works on *Crassostrea* species outside Indian waters. But it is unavoiadable to mention here and there about *Ostrea* species also for similarity and comparison. It is hoped that informations provided here will stimulate further interest in India where oyster resources are now known to be considerable and oyster farming is one of the priority areas in our efforts to produce more protein food from the sea and brackish water.

Substratum

Oysters grow equally well on a hard rocky bottom or semihard mud firm enough to support their weight. Shifting sand and mud appear to be unsuitable. Where oozy silt covers the bottom oysters suffer or die. (Bennet, 1946). With the exception of these extreme conditions oysters adapt themselves to a variety of bottoms. They thrive on shore rocks and underwater structures which are left exposed at low tides. Climate controls the survival since no oyster can survive several hours of exposure to below or above ambient temperature. Therefore very little settlement and growth is seen very near surface ; those clumps seen with dead oysters jutting above water level during low tides in estuarine zones or pillars of bridges represent the stock which have settled down when water level was adequate till seasonal vageries bring down water level leaving live oysters exposed to atmosphere resulting in the death of continuously exposed oysters.

In some estuaries the soft muddy bottom has been naturally improved by dead shell remains which give some firmness. Dead oyster shells dropping from clusters help the bed to grow horizontally or vertically. Galtsoff (1964) indicated an arbitrary scale from 0-10 to indicate the suitability of oyster bottom to oyster.

Habitat

It is difficult to state categorically that *Crassostrea* is an estuarine form just because it thrives well under estuarine conditions. It thrives equally well in sea water also. *C. madrasensis* lives and grows along Tuticorin coast (Tamil Nadu) and in salinities of 33-35‰ in the sea. It is known that Puerto Rico species, *C. rhizophorae* thrives at salinities 33-44‰ (Mattox, 1949). As stated by Fischer (1948) many so-called estuarine species have a wide ecological range and may endure salinities of oceanic water well. No doubt many oyster beds do especially well in estuarine waters bceause drills, starfishes and boring sponge cannot stand reduced salinities thereby indirectly helping oyster survival. Brackish water areas include all dynamically stable environment of lagoons, lakes etc. in which sea water diluted by freshwater is not necessarily influenced by tidal movements. On the other hand the estuarine environment is unstable. This makes it all the more difficult for expansive beds of oysters in the estuarine system. This accounts for the presence of several mounds of dead oyster shells found in the estuarine bar mouth region due to seasonal salinity variations.

Very often setting grounds of oyster spat are seen far above the spawning ground. In the distribution and abundance of oysters it can be said that sheltered bays often have a varied population of marine organisms as a result of wave action. Along open coasts, freshwater and sediment brought to the sea from rivers and streams are dispersed readily. Water movement is relatively weak, (Lewis, 1964) with the result that silt accumulates. A high percentage of animals with rock habitat are filter feeders. Water, high turbidity and the deposition of mud over the individuals tend to eliminate these animals from their habitats.

Light

Fischer (1948) has stated that little is known on the physiological influence of sunlight on oysters. Oyster spat settlement is greater in the darker under surface of objects than in areas which are less dark. It has also come to notice that in the discharge of spawns mature oysters react positively to darkness. Sun exposed oysters formed a thicker harder and more deeply cupped shell than those growing in the dark (Medcof and Needler, 1949).

Current

The areas with swift current are said to affect larval set and this is the reason why oyster spat are found at the bottom where the current action is weak than the surface where the action of current is greater in general. Tidal currents may carry away oyster larvae with it dispersing them in areas unsuitable for larval set or exposing them to plankton feeding animals.

Turbidity

Reports on the effects of turbidity on shellfishes in general are sometimes contradictory. It has been maintained that excessive turbidity inhibits the feeding mechanism thus restricting the growth. Lunz (1938) maintains that mortality of oysters is not increased by turbid conditions. Loosanoff and Tommers (1948) studied the effect of suspended solid material on feeding rate of oysters and concluded that oysters are vrey sensitive to suspended silt and there is an inverse proportion between concentration of sediment and rate of pumping. They state that even 0.1 g/l did seriously reduce the rate of pumping (to 40% of normal). If 3-4 g were added per litre of water only 4% of normal quantity of water was pumped. Very high concentration may inhibit pumping completely. Dead and dying oysters found in turbid waters invariably contain large amount of silt in their gills (Loosanoff, 1962). Korringa (1952) also found that silt and other substances producing turbidity, kaolin or chalk, at low concentration as 0.1 mg/l bring about reduction in water pumping. Oysters are normally not bothered by ordinary sediment but in some years of heavy spillway discharge there is 100% mortality. On the one hand there is a view that some turbidity in the water in which the larvae are living during metamorphosis may be desirable aginst U.V. light shielding these from U.V. radiation in shaded areas. There is a second view that the settling of turbid material to the bottom of a body of water prevents effective attachment of spat. Even 1 or 2 mm thick sediment is sufficient to prevent satisfactory oyster set (Galtsoff, 1964).

Loosanoff and Engel (1947) state that the rate of pumping may be influenced by the density of microorganisms present in the water. There are definite concentrations above which the density of microorganisms begin to interfere with the rate of pumping; not only it decreases but the oysters become sluggish. Dangerous quantities were 75,000/ml for smaller *Nitzchia* sp. and 2,00,000/ml for small *Chlorella* sp. This may be due to metabolic products of micro-organisms having inhibiting influence, probably caused by the bacteria which abound in such dense concentration and which live on organic excretion produced by the planktonic organisms. Water containing low concentration of small diatoms and dinoflagellates running over a bottom in a non-turbulent flow is ideal.

Water movements

Free exchange of water is essential for growth to remove gaseous and liquid metabolites and faeces and to provide oxygen and food. This is very essential for the dispersal of the oyster larvae in order to expand the beds. Estuaries are ideal in this regard. As the oyster closes its shell tightly underwater or exposed to air, many processes come to a standstill e.g., digestion. This process is said to be carried on anaerobically in closed oyster as long as glycogen reserves are available (Dugal, 1940). It is stated that *Crassostrea* is more hardy than *Ostrea* and can stand 24 days of exposure and live.

pH

In the case of C. *virginica* the pH of tidal estuary which forms the principal habitat must not fall below 6.75. The species does not reproduce successfully in waters where the pH remained above 9.00. The rate of pumping is normal at 7.75; in 6.75—7.00 vigorous first and then declines; at 6.5—4 decreases by 10% of normal.

Temperature

Almost all workers who have studied oysters from temperate waters have found that growth is confined to the periods when water temperatures are higher. Davis and Calabrese (1964) concluded that maximum growth results at a particular temperature since growth rate is affected by the type of food organisms available at different temperatures. Data on ecological range of adult oysters show that many oysters are killed by prolonged cold spells although *C. virginica* withstands —5°C. High temperature also causes death showing distress after 34°C. Functioning of adductor is weakened at 40°C and mortality results. The temperature regime affects the life of the oyster by controlling the rate of transport, feeding, respiration, growth, gonad formation and spawning. The oyster can adjust its pumping rate quickly to sharp and sudden changes in temperature (Loosanoff, 1950b). While it is recognised that temperature affects biological processes organismic response patterns to the natural environments are multidimensional. Most of the experimental studies devoted to the analysis of temperature effects have been conducted under simplified laboratory conditions. While such unifactorial approaches are useful there is need for the study of response to multivariable factor intensity patterns. Water temperature is known to influence the filtering activity i.e., the volume of water oyster pumps through its gills (Loosanoff, 1950a). From 28°—32°C the rate of pumping is as much as 37 L/hr. Normally 5—25 L/hr is pumped.

Reproduction tends to be confined to narrower thermal ranges than majority of other life processes.

The American oyster can feed and grow at much lower and higher temperature than are required for spawning. Once certain conditions such as appropriate physiological condition and nutritional demands are satisfied the time of reproduction is often decisively affected by temperature. Spawning of the oyster, *C. virginica* can be delayed by ripe or near ripe individuals being transferred to subnormal temperatures (Loosanoff and Davis, 1950). That is to conclude that temperature although high enough to permit gamete maturation is too low to induce spawning.

The time required for the larvae to reach the setting stage for *C. virginica* ranges in laboratory conditions from 10—12 days at 30°C—32°C to 36—40 days at 20°C (Davis and Calabrese, 1964). In the larvae of *O. edulis* the temperature range for satisfactory growth (70% or more) ranges at 27‰ salinity from 17.5°C—30°C. (Davis and Calabrese, 1964). Approximate setting time are 26 days at 17.5°C, 14 days at 20°C, from 8—12 days at 25°, 27.5°C and 30°C. According to Korringa (1941) larvae of *O. edulis* have a pelagic life of 6 days at 22°C—23°C, 9—10 days at 18°C—21°C and 13—14 days at 16°C—17°C. In the case of *C.gigas* the percentage of eggs developing to the shelled stages has a tendency to decrease sharply at temperature above the optimum (23°C—25°C) and decrease slowly at temperature below the optimum (Cahn 1950, Galtsoff 1964, Sato 1948, 1967). In *C. madrasensis* the spat settlement was achieved within 15-20 days at lower temperature range of 27°C—30°C although the optimum temperature was not decided upon (Nayar *et al.*, 1984). Medcof and Needler (1941), Thorson (1946), Korringa (1952) and Carriker (1946, 1951, 1961) give further information regarding thermal responses of lamellibranch larvae.

Enzymes required for molluscan larvae for digesting naked dinoflagellates are reported to be active at a lower temperature than are enzymes necessary to digest forms with more resistant cell walls. The temperature allowing maximum growth in larvae of *C. virginica* lies between 30°C—32.5°C at higher salinities and 27.5°C at low salinity of 7.5‰.

Salinity

Oysters like many other euryhaline organisms are able to live in seawater of wide range of salinity. Many have reported on adverse effects upon oysters of too low or too high salinities but statements are contradictory. The oyster can isolate itself from outside environment by closing its values tightly and survive adverse conditions provided they do not last indefinitely. Ranson (1943) noted damage below 13‰ when oodemic tissues are developed with vacuolisation of the epithelial cells and a sharp increase in leucocytes. Below 7‰ degeneration of tissue progresses. It has been observed that in Courtallayar river mouth, ranges below 10% slowly kill the Madras backwater oysters during north-east monsoon floods. In the Vaigai estuary at Athankarai the entire oyster population suffer total mortality when the salinity shoots up beyond 40‰ due to solar evaporation of the impounded water near the closed bar mouth. Unstable salinity regime characterises the tidal rivers and appears to be an important ecological factor.

Loosanoff (1950a) stated that sharp changes from low to high salinity can be withstood without physiological injuries and that it is tissue starvation due to prolonged low salinity exposure which leads to death. It is apparent that critical salinity values are to be determined for each area separately. Changes in salinity have not been known to induce spawning in the American or European oysters. Butler (1949) observed inhibition of gametogenesis in 90% of *C. virginica* in a low salinity area and attributed to inability of oysters to feed under low salinity conditions. Chestnut (1946) observes that in places where salinity is very low feeding often stops during low tide. Under tropical conditions of our coasts the temperature of the sea or the backwaters are maintained high throughout the year. Rao (1951) felt that a drop in density of water due to rains acts as a stimulating agent factor in the spawning of the Madras oyster in the east coast backwaters of South India.

Salinities between 5—8‰ are considered as an ecological boundary and referred to as 'horohalinicum'. This has been analysed by Khlebovich (1969) who found that the larvae of *C. virginica* tolerate salinity reductions down to 5-8‰. Adult requires higher salinities. In the case of *C. gigas* the formation of the larval shells is retarded in suboptimal salinities. Dupuy *et al.* (1977) reared the larvae of *C. virginica* and *C. gigas* to setting stage in 9-11 days at salinities 17.5‰—20.0‰. Hopkins (1937) found correlation between the periods of setting and periods of high salinity in *Crassostrea virginica* and that the larvae depended on a salinity of about 20.0‰ as a stimulus to develop the setting stage. In the case of *C. madrasensis*, Nagappan Nayar *et al.* (1984) found that the larvae reached the eyed stage from 13th to 15th day at salinities of 30.5‰. This was delayed further in higher salinities thereby indicating the role of salinity in larval development.

Larval food

Appreciation of nannoplankton organisms as food for marine larvae is increasing. That non-coloured flagellates like *Monas* may constitute a suitable food for oyster larvae under natural conditions is expressed by Imai and Hatanaka (1950). Needler (1941) and Blanco *et al.* (1951) have also expressed opinion that nannoplankton plays essential role in oyster larval development.

Fouling

It is of very great importance to understand the oyster bed associated organisms and the epifauna since they play a decisive role in the well being of oysters. Apart from abiotic factors which affect the oysters, heavy mortalities of oysters in the bed are caused by biological factors like predation, competition and disease outbreak.

The polychaete worm *Polydora* sp. is often mentioned as an agent causing damage. This worm selects the shell of the oyster as a habitat and diverts the energy of the oyster to shell secretion for countering the effect of mud blisters formed by the polychaetes. The shells are often honeycombed by the worms and become brittle. The infestation makes the animal vulnerable to secondary invasion by microorganisms. Similarly the boring sponge *Cliona* though not a true parasite excavates galleries in the calcareous shell of the oyster for shelter. This results in heavy mortality at times of heavy infestation by reduced resistence of oyster against pathogenic organisms (Old, 1941). In the case of English and Dutch oyster beds, the slipper limpet *Crepidula fornicata* is considered a serious pest (Korringa, 1952) by occupying space meant for young oysters.

Pollution

Pollution plays an important role in the ecology of oyster beds from the biological point of view. In coastal waters contamination of water by domestic sewage and trade waters are common. The sewage covers the bottom with a sludge which smothers the oyster bed affecting the oxygen content of the water and greatly increasing the bacterial load of the water. The degree of pollution is determined by *Escherichia coli* found in the water. Where the MPN is in excess of 70 per ml the area is unsafe. Similarly the disposal of radio-active waste in the sea presents a new threat. Excess heavy metals in water like Mercury and Zinc tend to get accumulated in oyster flesh and beyond certain level prove to be lethal. Sulphite waste liquor of pulp mills has caused damage. Odlaug (1949) found that in 1,000 p.m. concentration the oysters suffered greatly due to perhaps impairment of mucus feeding sheets and also gill lamellae forming indentations (Mackernan, Tartar and Tollefson, 1949). Oysters developing abnormal fibres in the adductor muscle (Galtsoff, Chipman, Engel and Calderwood, 1947). As a sedentary animal not capable of locomotion after setting the oyster is vulnerable to environmental changes thus affecting its equilibrium or steady state. Factors of environment act jointly and the combined action of several factors produce a far greater effect than that by a single factor. A thorough understanding of all these is essential in the ecological studies of oyster bed. Some pollutants contain highly toxic substances and cause mortalities among marine populations. Others are not so toxic to have lethal effects on adults but decrease the rate of survival of their larvae. Davis (1961) found DDT to be one of the most toxic with 0.05 ppm causing 90% mortality of oyster larvae. The normal ecological environment may be so changed, that some planktonic organisms useful to shellfish as food disappear and are replaced by microorganisms not useful but even harmful.

Diseases and parasites

Extensive studies have been made on the mortality of oysters in natural beds caused by the pathogenic fungus *Parkinsus marinus* (Dermo), the haplosoridians *Minchinia nelsoni* (MSX) and *M. coastalis* (SSO) in the case of *C. virginica*. Similarly in France and other European coastal beds *Marteilia refringens* causes large-scale mortalities. In India, so far, such pathogens do not seem to occur ; but a suspected case of Dermo in *C. madrasensis* is being carefully studied. Mahadevan (1980) has catalogued most of the prominent oyster diseases reported from all over the world. Apart from MSX, SSO and Dermo diseases the list includes *Ostracobiabe implexa* (*C. virginica*) *Myotomus ostrearum* (*C. virginica, C. gryphoides*), *Nocardia*, (*O. edulis*) *Nematopsis ostrearum* (*C. virginica*) *Hexamita nelsoni* (*O. edulis, O. lurida*), *Sphaenophyra*, (*C. virginica*) *Nosema dollfusi* (*C. virginica*), *Orchitophyra stellarum* (*C. virginica*) and *Ancistrosoma pelseneeri* (*C. virginica*) as of considerable importance as disease causing agents. Crustacean parasites like *Mytilicola intestinalis* in *C. gigas* and helminths (cestodes and trematodes) also cause debility to the oysters.

Predation is next to disease causing agents in eliminating oysters from beds. Crabs in the oyster beds crack off the edges of oyster shells which leads to

death of oysters ; particularly the spat are more vulnerable. Predatory gastropods known as drills play havoc to oyster population. *Urosalpinx cinerea* in Atlantic coast is a classic example. In *C. madrasensis* beds the gastropod *Cymatium cingulatum* destroys the spat and adults. Although *Thais* and *Rapana* are also known to be dangerous their threat is much less to *C. madrasensis*. The havoc played by these differs from one geopgraphical area to the other. Control measures have been evolved and followed with certain amount of success but the problem always continues in the natural beds resulting in losses of natural population.

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3

OYSTER RESOURCES OF INDIA

S. MAHADEVAN[1]

Of the 8 species of *Crassostrea* listed by Awati and Rai (1931) only *C. madrasensis* (Preston), *C. gryphoides* (Newton & Smith), *C. discoidea* (Gould) and *C. cucullata* are known to be economically and commercially important so far as India is concerned. Following Stenzel (1971) who recognised *Saccostrea* as one of the eight genera of living oysters the tuberculated species, *Crassostrea cucullata* is now designated as *Saccostrea cucullata* (Born). It has now been reported by Rao (personal communication) that it is *C. rivularis* (Gould) that occurs all along the Gujarat Coast and in some regions of Maharashtra coast. Judging from the commonness and distributional abundance of these 4 species under the two genera found in India it can be reasonably stated that *C. madrasensis* is the native oyster of India and *C. gryphoides* and *C. rivularis* may be designated as west coast oysters for the purpose of convenience.

Saccostrea cucullata is purely a marine form. Although it occurs all along the coast of India nowhere is it found to form prolific oyster beds unlike the other three species. *Crassostrea* spp. occurring in India are euryhaline and are found in estuaries, backwaters and open sea coastal shallows. These are found either as large aggregations where the substratum for their settlement, survival and growth is ideal or as patchy aggregations on submerged objects suitable for attachment. Dense occurrence is seen in the proximity of bar mouth of estuaries possessing gritty bottom or otherwise conducive for their settlement. Both the west and east coasts of India possess many productive areas and a few moderately productive areas of these species. It is of particular interest to mention that *C. gryphoides* and *C. rivularis* are restricted to the northwest coast regions and not reported so far anywhere in the east coast.

From the resources point of view, Tamil Nadu and Kerala are rich in *C. madrasensis*, Maharashtra and Goa in *C. gryphoides* and Gujarat in *C. rivularis*.

Unlike other molluscs like clam and mussel most of the edible oyster beds are clumsy formations and mostly dispartate, The horizontal and vertical aggregations of individuals build up one over the other cemented together, thus giving rise to mounds. The 'oyster reef' so formed usually consists of different size and year groups of oysters with a considerable percentage of dead shells. This makes population estimation difficult. The locations of many beds are also not so easy of approach. It is therefore not surprising that our understanding of the actual extent of the oyster beds in each state, population density, magnitude of annual recruitment etc. is still imperfect. Creation of a special task force of scientists to assess the oyster resources potentiality of each state is therefore very necessary on the model of what has been done for the oyster beds in York river, James river, Plankatank, Rappahanrock and Wicomico rivers in Virginia State of the United states of America. The information so gathered by us will be of great help in the future exploitation of oysters which otherwise is at present spasmodic and underexploited.

The present chapter gives an account of what little information that is available on the oyster resources on an all India basis.

Crassostrea madrasensis (Preston)

West Bengal

Very little is known about the resources along West Bengal coast.

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001.

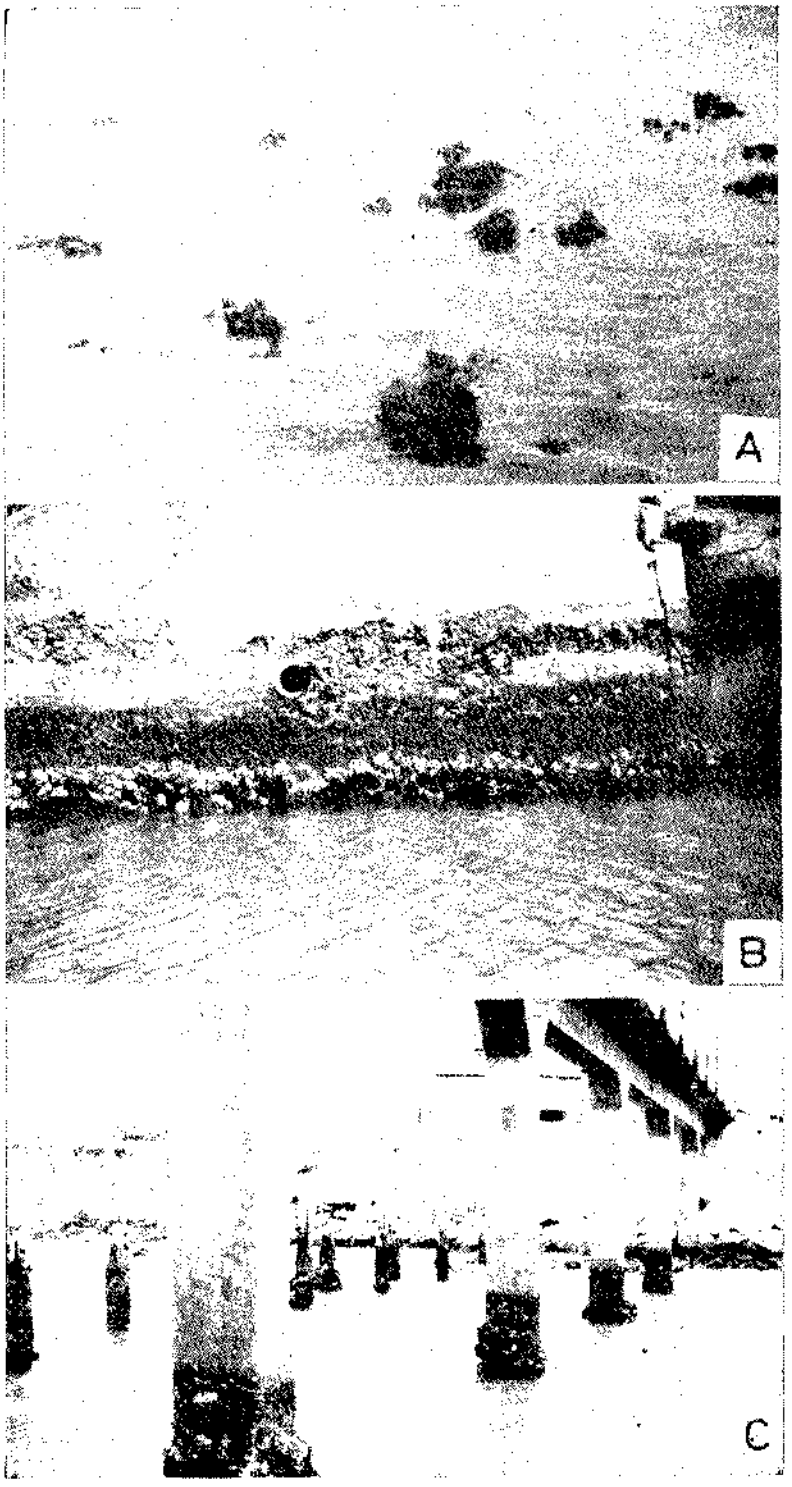

PLATE I A. A bed of *Crassostrea madrasensis* in Pulicat Lake partially exposed at low tide. B. *C. madrasensis* settled and growing on the embankment of a bridge at Tuticorin. C. The oysters growing attached to the pillars of an abandoned bridge at Karapad, Tuticorin.

Orissa

The state has a coastline of 480 km. and brackish waterspread of 0.4128 million ha. The only area where oyster beds are known to exist is the Bahuda river estuary near Sonapur. An approximate area of 5 ha. of the river bed close to the bar mouth contains 3 distinct beds. The oyster population was estimated in this area as 2,500,000.

Andhra

The state has a coastline of 982 km. with 0.566 million ha. of brackish water. Oyster beds are reported in Sarada estuary and near Waltair. In Bhimunipatnam, the oyster beds are subjected to annual depredation due to fresh water influx in the area, making it difficult to assess the actual density of the population. In Upputeru canal banks 2.25 hectare is reported to contain millions of oysters. Although they are reported from Godavari and Krishna estuaries, the population density is very thin. Another bed of considerable extent is known from Gokulapalli. Here oysters are regularly exploited.

Tamil Nadu

The state has 1,000 km. long coastline and 0.17 million ha. of brackish water. Oyster resources are rich compared to other states. Naturally the oyster beds in this state have been better studied than in other states. Pulicat backwaters (Lake) is famous for the extensive oyster beds. Areas like Chinnaparaval bed, Karimanal bed, Dhonirevu, Moosamani, Kottakuppam, Sathankuppam, Kondurpatnam and Dugarajapatnam are well known for the oyster beds. The extent of each bed varies from 4 hectares to 10 hectares. The oysters in these beds are characteristically elongate, long, narrow or subspatulate in form. They tend to segregate into clusters varying 3 to 4 individuals in each. They are sparsely scattered over the bottom, shallower region showing greater density. Although the entire lake is subject to the influx of flood water during the rainy season large scale mortality of oysters does not take place. It is estimated that the total oyster population in the lake is about 11 million. Regular fishing is reported from one or two beds like Karimanal. The Courtallayar estuary at Ennore and Adyar estuary at Madras possess oyster beds to an extent of 50 hectares. The population here is often affected by flood water admixture. Regular exploitation by fisherfolk goes on.

In Killai backwaters (near Cuddalore) at Mudasodai and Chinnavaykal there are 3 beds containing oysters. Exploitation is limited. At Muthupet swamp, patchy settlement is noticed. The Vaigai estuary at Athankarai possesses two hectare area of oyster beds having about 3.5 million oysters. Exploitation is not done. These oysters suffer periodically due to abnormal salinity of the water in the estuary on account of solar evaporation of the impounded water when the bar mouth remains closed during summer. At Tuticorin there are 3 tidal inlet beds with a total area of 20 hectares having a population of 1.5 million oysters. In Tamparaparni estuary at Pinnakayal there is a bed of 2.5 hectares in the upriver region. The oysters here are sparsely distributed. Exploitation in this area is not reported (Pl. I a-c).

Kerala

Kerala has a coastline of 560 km. and a backwater area of 0.33 million ha. Oysters are found in 5 hectare area in Ashtamudi Lake. Dalavapuram, Kavanadu, Kuripuzha, Karichal, Panathura, Thirumullavaram to Quilon are all areas where oysters are found sparsely distributed and exploited. Anchengo backwaters also contains good population of oysters. In Vembanad Lake there are distinct oyster beds but the density of population is thin. Apart from this Neendakarai, Kannamali, Maruvekkadu, Punnappara, Thottapally, Chaliyar estuary, Beypore, Azhikode, Elathur, Calicut, Tellicherry and Cannanore also possess oyster beds of limited extent. These are exploited by the local fisherfolk. The extent of oyster beds in these areas mentioned above has not been determined so far.

Karnataka

The coastline is limited but the state abounds in many estuaries. Of these, Nethravathi, Sharavathi and Kali river estuary possess oyster beds of limited extent ranging from 1 ha. to 5 ha. In addition, Mulky river estuary, estuaries at Uppunda, Bhatkal, Venkatpur and Coondapoor show oyster beds of some extent. These beds are being exploited regularly.

Apart from the utilization of live oyster meat as food the dead shells are collected for industrial purposes. Mining of subfossil deposits by lessees carried out in many estuaries like Kali river, Athankarai and Bahuda river yield nearly 15,000 t of oyster shells annually.

Crassostrea gryphoides (Newton and Smith)

Maharashtra

The state has a coastline of 720 km and brackish waterspread of 0.1214 million ha. The oyster beds are not very extensive as in the case of *C. madrasensis*

but unlike the latter wherever beds of *C. gryphoides* are located exploitation is regularly done.

Dahanu Creek, Boiser, Satpuri, Palghar, Kelwa, Malad, Navapur, Utsali, Dahisar, Mahim Creek, Alibag, Purnagad, Ratnagiri, Jaytapur, Malwan, Worli, Versova, Marve, Gobbunder, Cuff Parade, Bandra, Madh, Bhate Bunder, are all areas where beds are found and population harvested periodically.

In many places like Utsali, Navapur and Kelwa bottom culture is done traditionally by fisherfolk. (Alagarswami and Narasimham 1973, Silas et al. 1982) Unfortunately details of annual production from natural beds and by culture are lacking.

Goa

The territory has a coastline of 153 km. Oyster settlement is reported from Ribander, Siolim and Curca. Extent of grounds and magnitude of annual production is not available.

Crassostrea rivularis (Gould)

Gujarat

The state has a coastline of 1,663 km and brackish waterspread of 0.4189 million ha. The oyster species occuring here has been reported to be *C. discoidea* by Awati and Rai (1931). As already mentioned Rao considers that it is *C. rivularis* which is distributed in Maharashtra and Gujarat coasts.

The oysters are found in intertidal hard grounds and in muddy creeks. Aramra, Poshetra, Port Okha, Porbander, Sikka, Gagwa creek, Singach creek, Beet Kada, Khanara creek, Laku Point, Gomati creek (Dwarka), Harsad, Navibander (Medha creek), Balapur, Azad island are all places where there are settlements. Exploitation is done regularly. But data on extent of beds, population density and annual production are wanting.

Maharashtra

In Mahim, Ratnagiri and Jaytapur areas also this species is found along with *C. gryphoides.*

Saccostrea cucullata (Born)

This species is found all along the Indian coast in shallow areas wherever the substratum is rocky or very hard. But nowhere does it form beds large enough for exploitation. Nevertheless, those found along Maharastra and Gujarat coast are numerically large, collected and utilized as food. The quantity exploited is insignificant when compared to other three species.

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4

OYSTER RESOURCES OF ATHANKARAI ESTUARY, SOUTHEAST COAST OF INDIA

K. SATYANARAYANA RAO[1], D. SIVALINGAM[2], P. N. RADHAKRISHNAN NAIR[3] AND K. A. UNNITHAN[4]

INTRODUCTION

The oyster *Crassostrea madrasensis* is distributed at several places along the east and southwest coasts of India (Rao 1969, Alagarswami and Narasimham 1973) and has good economic potential but so far resources survey of the shellfish populations has not been carried out in any area. Estimates of the standing stocks are essential for planning exploitation of the oyster resources. A precise knowledge of the natural stocks and the ecological conditions in which they abound is especially necessary to plan and conduct oyster culture which is essential for large scale exploitation. Beds of oysters belonging to the species *Crassostrea madrasensis* are present in Athankarai Estuary at Athankarai near Mandapam Camp on the southeast coast of India. The highly nutritious shellfish resources are not exploited by people of the area for food. Only occasionally oyster shells are collected and burnt in small kilns and converted into lime. The oyster resources of the estuary have been surveyed. The general features of the Athankarai estuary, hydrological conditions, ecologically associated fauna and flora, distribution and magnitude of standing stocks of oysters, and seasonal changes in meat of oysters have been studied and the results are presented here.

MATERIAL AND METHODS

The occurrence of oyster beds in the estuary was throughly searched by preliminary inspection and diving in the estuary. The oysters occur in patches or larger-sized formations. The shape of each patch was determined by noting its length and breadth at intervals of 1 metre (Pl. I A-C), plotting the measurements on a graph sheet and the shape and area were found out. For determining the density of oysters, all the oysters present throughout the height of the patch in $\frac{1}{4}$ metre square area were counted and the number of oysters per sq. m. was taken as the density of the oysters. Counts of oysters were made in two to four $\frac{1}{4}$ metre square quadrats and the averages of the same were taken as the density in the patch or formation. From the density, the total number of oysters present in each patch or formation was calculated. The total weight of oysters (shell on) present in $\frac{1}{4}$ metre square area was determined and from the average weight of oysters in 2 to 4 half metre square quadrats in each patch, the oyster biomass in the total area of each patch was calculated. The total meat weight of oysters in the various patches was also similarly estimated. Data on the size, weight, meat weight and stages of maturity of oysters as also epiflora and epifauna were recorded. Oyster shell and concrete piece spat collectors were kept suspended from horizontal bamboo poles supported on casurina poles in the different parts of the estuary viz., near the mouth, middle and upper portions of the estuary to determine the intensity of spatfall. Data were also collected on the temperature, salinity and dissolved oxygen content of water over oyster beds in the estuary. The resources survey was conducted between July and September, 1975.

A random sample of ten oysters of *Crassostrea madrasensis* was collected every month from Athankarai Estuary from August to July, cleaned and weighed. The oysters were shucked with a chisel and weight of meat of the oysters determined. The weight of meat of an oyster expressed as a percentage of total weight of the oyster is the percentage edibility. Then the stage of maturity of the oysters was determined by microscopic examination of the gonads. Oysters in various stages of maturity viz., immature, maturing, mature, partly spawned, spent, hermaphrodite, indeterminate and recovering were recorded in all the periods of the year. The entire soft parts of each oyster were uniformly pressed between two pieces of Whatman No. 42 filter paper and adhering mantle fluid removed. The meat of the oysters was weighed to constant weight in a hot air oven at 100°C. Oven drying method was adopted instead of drying over sulphuric acid using suction pump as the material retained some moisture even after prolonged drying in the latter method. The total solids content was determined calculating

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001.
[2] *Present address :* CMFRI, Regional Centre, Mandapam Camp-623 520.
[3] *Present address :* CMFRI, Research Centre, Vizhinjam-695 521.
[4] *Present address :* CMFRI, Cochin-682 031.

the dry weight of meat of each oyster as percentage of the wet weight of the oyster. The water content was similarly calculated from the difference between the wet and dry weights of meat of each oyster. The dried oyster meats were homogenized and the lipid content was estimated by extracting 100 mg of homogenized meat in a soxhlet apparatus with ethyl either and drying the ether extract.

from Mandapam Camp. The Athankarai Estuary meanders into this region from west running parallel and close to the National Highway 49 and curves to a northern course when about 1.82 km from the imbochure (Fig. 1). The width of the estuarine system ranges from 130 m (upper reaches) to 356 m very near to the estuary mouth. The estuary gets cut off from the sea in the period from April to June due to the forma-

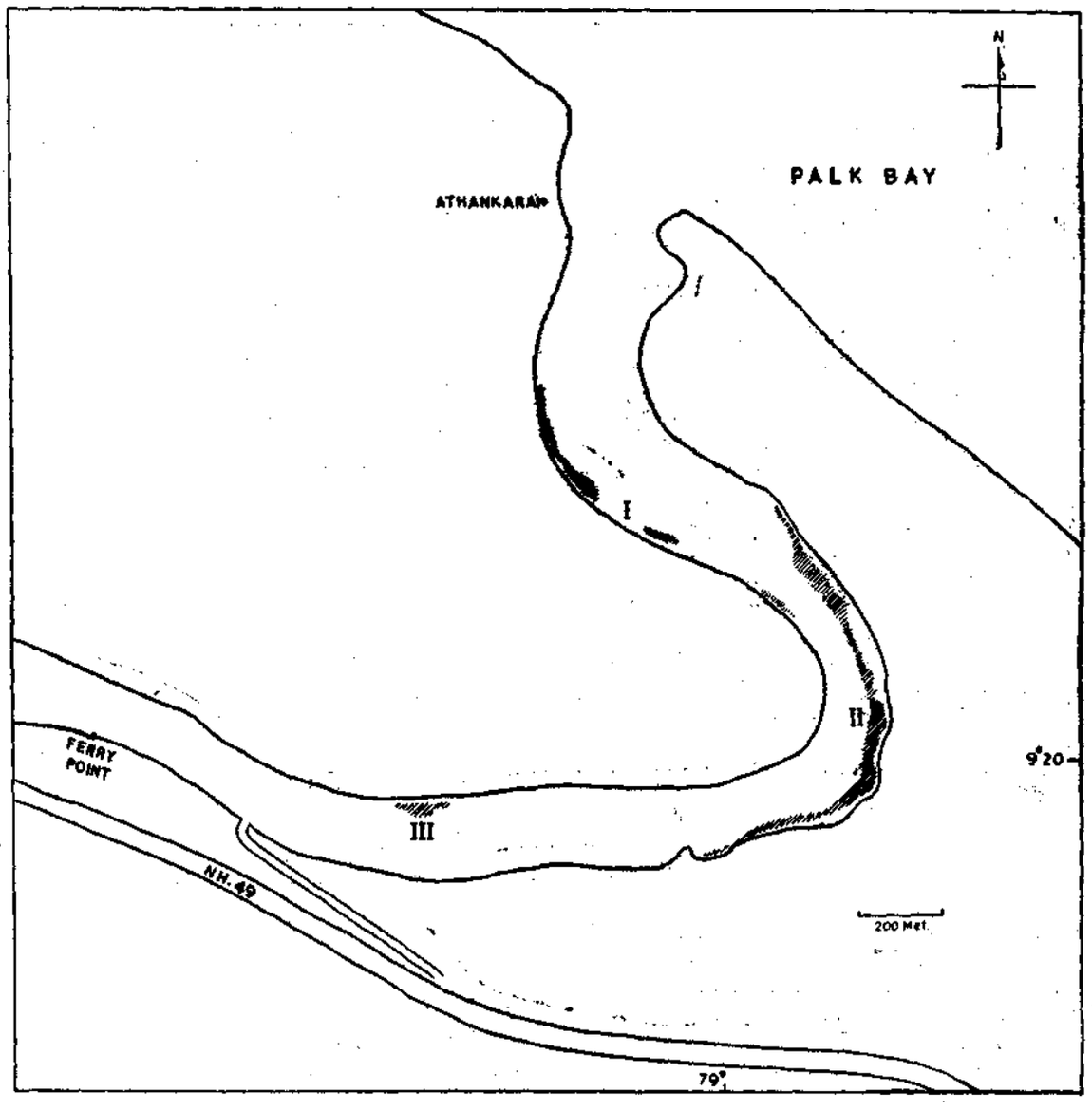

Fig. 1. Map showing location of the I, II and III oyster beds in Athankarai Estuary.

General Features of Athankarai Estuary

River Vaigai confluences with the sea on the Palk Bay side (Durve and Alagarswami, 1964) on the eastern side of Athankarai village which is situated at Lat. 9°.20′ N and Long. 79° E and is at a distance of 25 km

tion of sand bar which opens when the river receives freshets on account of southwest monsoon rains. In some years the sand bar is formed early in January itself. The estuary is shallow with a maximum depth of 3 m in the middle while near the banks it varies from 1 to 2 m. Due to the estuary becoming inundated

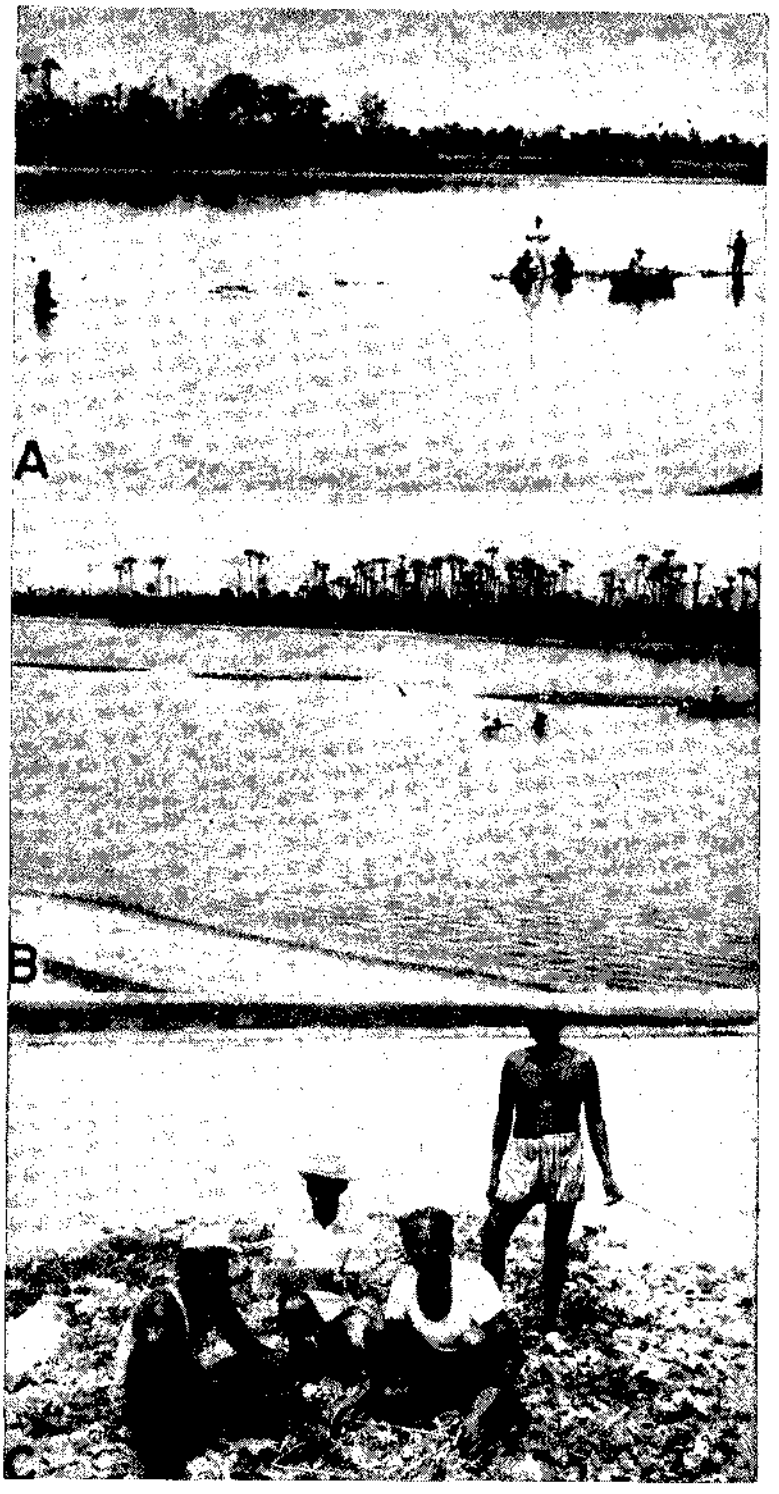

PLATE I. A and B. Recording of measurements of oysters beds in Athankarai estuary. C. Counting of oysters from an oyster patch exposed at low tide, in a $\frac{1}{4}$ metre square quadrat.

during the northeast monsoon period October-December, the depth near the banks increases to 3 m and that at the middle to 4 m.

The bottom of the estuary is throughout muddy with an admixture of sand and a large amount of organic detritus. The mud is grey in colour. The seagrasses *Cymodocea serrulata, C. isoetifolia* and *Halophila ovalis* are found growing in the estuary especially near the banks. A variety of fishes such as *Himantura alcockii, Amphotistius imbricatus, Thrissocles mystax, Tachysurus thalassinus, T. caelatus, Mugil cephalus, M. parsia, Apogon aureus, Therapon spp., Leiognathus jonesi, Gerres filamentosus, Scatophagus argus* and *Tripterygion fasciatum* and three prawns *Penaeus indicus, Penaeus semisulcatus* and *Metapenaeus burkenroadi* are found in the estuary. The clam *Meretrix casta* and the prosobranch gastropod *Cerithidea fluviatilis* also occur, the former forming beds in the middle and upper parts of the estuary. The salinity of the estuary shows a steep fall to 17.83‰ in some months but *Crassostrea madrasensis* which thrives well in brackish waters survives in the low salinity conditions.

Hydrology of the Estuary

Recorded data on the temperature, salinity and dissolved oxygen content of Athankarai estuarine water over the three oyster beds in the period July, 1975 to September, 1976 are given in Table 1. It may be seen from the Table that water temperature of the estuary varied from a minimum of 27.5°C recorded in December, 1975 to a maximum of 34.5°C observed in August, 1975. In the period December, 1975 to February, 1976 coinciding with the monsoon and winter periods the temperatures were low, being 27.5°C to 29.2°C and in the other months the temperature was higher. The water temperature at stations I and II did not show distinct difference but those at Station III were slightly lower than at station I except in September, October and December, 1975 and January, February, August and September, 1976. Salinity varied from 17.83‰ (April, 1976) to 71.21‰ (September, 1976). Except in July, 1975 and January, February, July and August 1976, in all other months the salinity values were slightly higher at Station III than at Station I. The salinity values at Station II were same or slightly different from those at Station I. The estuary was not connected with the sea from the last week of January, 1976. The highest salinities 48.43—53.12‰ recorded in August, 1976 and 62.43—71.21‰ in September, 1976 were due to excessive evaporation of estuary water. The dissolved oxygen concentration showed a range of 0.86 ml/l in July, 1976 to 8.74 ml/l in March, 1976. High content of dissolved oxygen 4.89—8.74 ml/l was noted in all the three stations in February—March 1976. Clearly marked differences in the dissolved oxygen concentrations between the three stations of the estuary were, however, not discernible.

Table 1. *Water temperature, salinity and dissolved oxygen in the period July, 1975 to September, 1976 at three stations where the three oyster beds are situated in Athankarai Estuary.*

	Temperature° C Stations			Salinity (‰) Stations			Dissolved oxygen (ml/l) Stations		
	I	II	III	I	II	III	I	II	III
July, 1975	32.5	31.0	30.0	21.35	20.10	19.36	4.74	4.35	4.06
August	34.5	34.5	34.2	33.23	33.00	32.52	3.77	4.34	6.47
September	33.0	33.2	33.2	34.26	35.10	36.24	3.46	4.06	5.32
October	30.7	31.0	30.7	32.40	33.45	33.58	5.16	5.18	5.36
November	30.0	29.7	29.5	30.88	31.17	31.11	2.27	3.76	4.12
December	27.5	27.7	27.7	25.95	24.74	25.25	4.60	4.18	4.77
January, 1976	28.5	28.5	28.7	23.43	22.28	23.27	4.52	4.55	4.79
February	29.2	29.2	29.2	24.38	23.56	24.01	5.94	4.89	7.09
March	31.5	31.0	31.0	27.81	27.81	29.80	8.74	8.58	6.72
April	31.5	31.0	31.0	17.83	17.96	18.30	4.86	5.42	6.02
May	32.7	30.4	30.7	24.29	24.28	25.98	4.54	4.49	3.87
June	32.9	32.0	32.0	30.29	30.29	35.84	3.71	4.31	3.33
July	28.9	28.7	26.5	37.18	37.18	34.20	4.04	4.67	0.86
August	28.7	29.5	29.7	62.99	62.43	71.21	2.44	2.44	2.68
September	27.8	28.2	28.5	62.99	62.43	71.21	2.44	2.44	2.54

Distribution of Oyster Beds in the Estuary

The survey revealed that oysters belonging to a single species, *Crassostrea madrasensis* (Preston) along with stray individuals of *Saccostrea cucullata* occur in the estuary. There are three oysters beds in the estuary, each bed consisting of a number of patches and larger formations with clusters of oysters forming large irregular structures (Fig. 1). The first oyster bed is found along the western bank of the estuary commencing at a distance of 0.72 km from the mouth of the estuary and extending for a distance of 0.93 km. The second oyster bed is present along the eastern bank in the upper portion of the estuary. The first formation of this bed is located 1.03 km from the mouth of estuary and the bed extends over a distance of 1.28 km. The third bed is present still further up in the upper portionof the estuary near the northern bank 2.94 km from the mouth of the estuary and it extends over a limited distance of 120 m. There are no oyster beds near or west of the Ferry Point which is 3.75 km. from the mouth of the estuary.

A. I Oyster Bed

The oyster bed consists of 41 patches and formations which are close to the western bank of the estuary at a distance of 1.5 m to 3 m from it (Fig. 1). The patches and formations are in a row, one behind the other, most of them at intervals of less than a metre or 1 to 37 metres from one another. Patch 29 and formation 34 are 134 m and 124 m respectively from the previous patches. They are low in the case of beds 1 and 2 and others are raised to a height of 0.2 m to 1.4 m from the bottom of the estuary. Most of the oyster patches of this bed are small sized with an area of 0.25 sq. m. Formations 5, 6, 30, 31 and 34 are larger, their area varying between 117 sq. m. and 176.5 sq. m. The estimated total area of all the 41 patches and formations of the bed is 1,366.84 sq.m (Table 2). The shape of patches 1 to 4, 16, 22, 32, 35, 37, 39 and 41 is approximately round or cordate while others are irregular. In this bed the density of oysters varied in different patches. In patches 1 to 4, 8, 12, 14, 27 and 28, the density of oysters is only 56 to 96 per sq. m. High density of 300-520 oysters per sq.m. was observed in patches 19, 29, 32 and 35 and formations 30, 31, and 34. In the other twenty four patches the oyster densities ranged from 105 to 288 per sq.m. The number of oysters per patch varied from 14 in patch 1 to 62, 930 in formation 31. The estimated total number of oysters present in the bed is 3, 44, 116. The estimated total or absolute biomass of oysters per patch ranged from 2.09 kg in patch 1 to 9, 691.22 kg. in formation 31 with an average of 1,183.09 per patch and the estimated over all total biomass of oysters in the bed was 48,506.89 kg. (Table 2). The estimated total meat weight of oyster per patch in the bed varied from 0.13 kg. in patch 1 to 358.70 kg in formation 31 with an average of 48.43 kg. for the patch and the estimated total meat weight of oysters in the bed was 1,985.79 kg.

Biological aspects of oysters

In this bed the oysters ranged from 31 mm in size with average size of 100 mm (for 629 oysters). 83% of the oysters were in the size range 61-140 mm (Fig. 2)

Table 2. *The estimated area of the three oyster beds in Athankarai Estuary and the estimated number of oysters, biomass of oysters and meat weight of oysters in the three beds*

Serial number of oyster beds	Estimated area of the oyster beds (sq. m)	Estimated number of oysters in the beds	Estimted biomass of oysters in the beds (kg)	Estimated meat weight of oysters in the beds (kg)
I Bed	1,366.84	3,44,166	48,506.89	1,985.79
II Bed	13,692.85	30,50,533	3,25,164.92	11,111.72
III Bed	562.22	77,923	15,249.48	576.59

Oysters below 50 mm in size formed only 3.7%. The weight of the oysters ranged from 29 gm to 232 gm with an average of 112.6 gm and meat weight from 1 gm to 15.6 gm with an average value of 4.7 m. Oysters setting on oyster shell cultch and 8-24 on concrete piece cultch in the period October to March. On the other hand only stray spat set on cultch kept suspended near the mouth of the estuary.

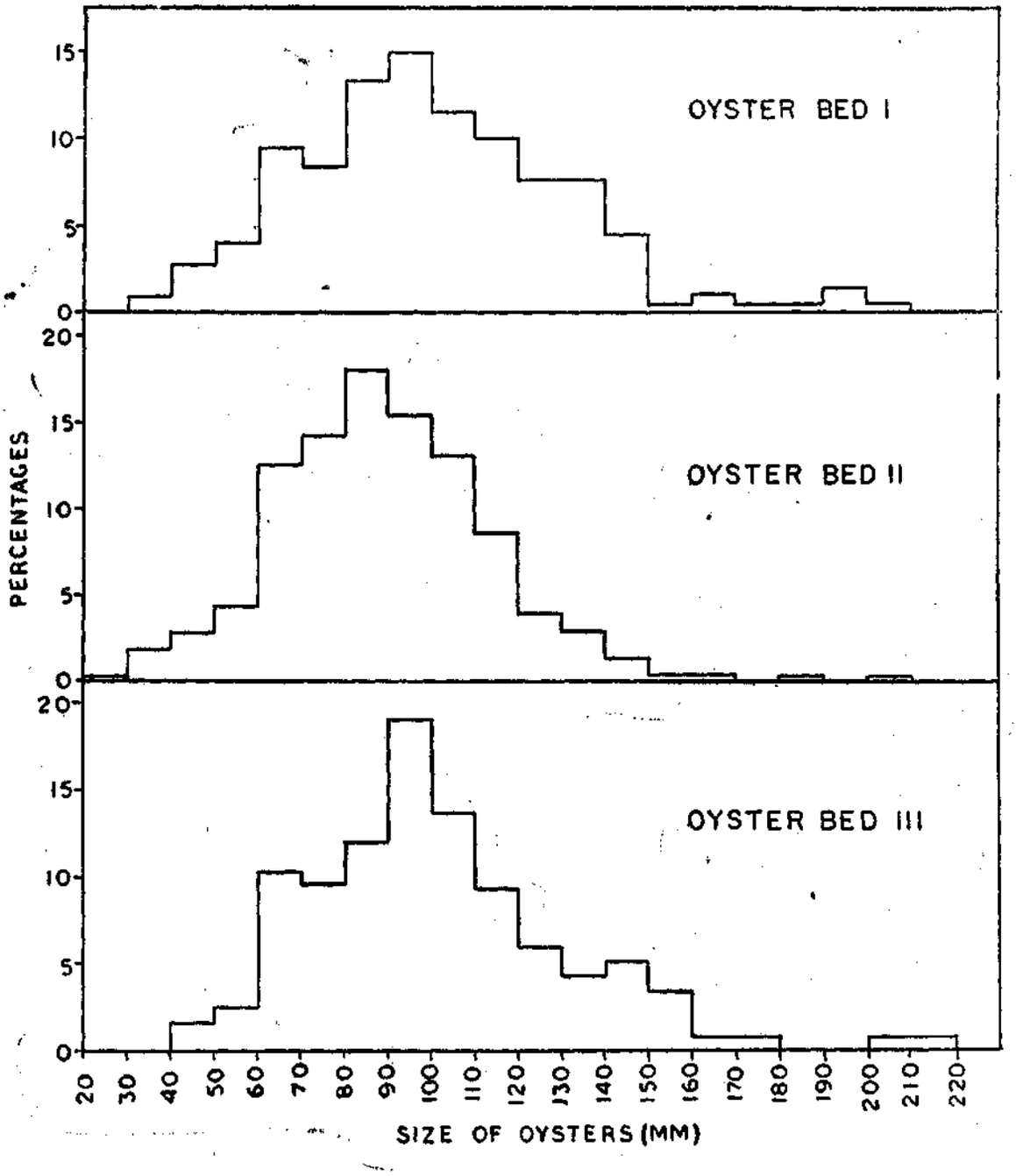

Fig. 2. Percentage frequency of oysters in I, II and III oyster beds in Athankarai Estuary.

of both sexes were in immature, maturing or sexually ripe stage. The meat of the ripe oysters was thicker than that of immature and maturing ones. There was good spatfall in this area with about 6 to 12 oyster spat

Ecological Conditions

The algae *Enteromorpha* sp. and *Polysiphonia* sp. were common and formed a felt-like covering on the outer surface of valves of oysters but they did not cause any

significant damage to the oyster valves as they were strong and unaffected. Blue-green algae also occured on the valves of oysters in density. The weaving mussel *Modiolus undulata* and the barnacle *Balanus amphitrite communis* were present in small numbers as epifauna. The boring polychaete *Polydora ciliata* was noticed in some oysters. Only a few individuals of the oyster drill (*Thais rudolphi*) were recorded on the oyster bed. Hermit crabs (*Pagurus* sp.) were recorded in small numbers and these were predators of oyster spat and oysterlings. Stray individuals of the crabs *Carpilodes margaritatus*, *Xanthe* (*Leptodius*) *euglyptus* and *Scylla serrata* were seen on the oyster bed. There was heavy settlement of barnacles on the poles and cultch used for studying spatfall.

B. II Oyster Bed

51 oyster patches and formations constitute this bed. These are located 4-11 m from eastern bank as in the case of formations 1, 3, 15, 22, 35 to 37, 45 and 48 and patches 2, 38, 39 to 44, 46 and 47 are 20-45 m from the eastern bank as in the case of formations like 6, 12, 20, 21, 23, 31, 32 and 35. The oyster patches of this bed had a height of 0.2 m to 1.75 m from the bottom, the patches farthest away from the bank having a height of 1 to 1.75 m from the bottom. In this bed thirty patches *viz.*, 2, 9, 10, 13, 14, 16 to 19, 21, 24, 26 to 29, 31, 33, 34, 38 to 44, 46, 47 and 49 to 51 were small, measuring 3.25 sq. m. to 85.37 sq. m. in area. Eighteen formations 1, 3, 4, 5, 7, 11, 12, 15, 20, 22, 25, 30, 32, 35 to 37, 45 and 48 were larger with an area ranging from 113.12 sq. m. to 621.62 sq. m. Oyster formations 6, 8 and 23 were very extensive with areas of 2,207.37 sq. m. 2,705.62 sq. m and 2,208.37 sq. m. respectively. The estimated total area of the bed is 13,692.85 sq. m. (Table 1). The smallest patches were round, oval or cordate while medium size ones were mostly irregular. Formations 6, 8 and 23 were very long and broad with length ranging between 188.5 m 231.5 m and width ranging between 24 m and 29.5 m.

In this bed the density of oysters varied from 124 to 608 per sq. m. In some areas 1 to 5, oysters in high densities of 340 to 480 per sq. m. were recorded. In the remaining localities, the density of oysters ranged from 108 to 272 per sq. m. The estimated total number of oysters present per formation in the bed varied from 598 in patch 44 to 5,43,829 in formation 8 which is also the largest formation in the estuary. The estimated total number of oysters present in this bed is 30,50,533. The estimated total biomass of oysters per formation in this bed ranged from 78.45 kg in patch 44 to 58,026 kg in formation 8 with an average of 6,375.78 kg for a formation and the estimated over all total biomass of oysters in this bed was 3,25,164.92 kg. (Table 2). The estimated total meat weight of oysters varied from 3.22 kg to 1,731.13 kg with an average meat weight of 217.87 kg and the estimated overall meat weight of oysters in the bed was 11,111.72 kg.

Biological aspects of oysters

The oysters in this bed showed a size range of 24 mm to 206 mm with an average of 99 mm for 835 oysters. 88.3% of oysters were in the size range of 61-140 mm. Oysters below 50 mm in size formed 4.9% which is higher, compared to that in the previous bed. The weight of oysters varied from 22 gm to 320 gm with an average weight of 106.5 gm and the meat weight of oysters from 1 gm to 8 gm with an average of 4.1 gm. In addition to male and female oysters, hermaphrodite oysters showing sex reversal were observed. Immature, maturing, ripe, partly spawned and spent stages were recorded. The meat of the maturing oysters was moderately thick, that of ripe ones was thick and cream coloured and that of partially spawned and spent ones thin and watery. Good oyster spatfall was observed with 3 to 4 spat setting on oyster shell cultch and 16 to 21 spat on concrete cultch.

Ecological Conditions

Enteromorpha sp., *Polysiphonia* sp. and Myxophyceae were found as epifauna of oysters. In addition *Padina* sp. was also found in some places. Barnacles (*B. a. communis*) occurred in small numbers. *Modiolus undulata* were very common on the oysters. Only a few oyster drills *Thais rudolphi* were found on the oyster beds. The polychaetes *Marphysa gravelyi*, *Eunice* sp. and *Polynoe* sp. were found in the crevices between oysters. The boring polychaete *Polydora ciliata* was seen in some cases causing damage to valves of oysters. Sponges (*Hyatella cribriformis*), amphipods and alpheids were common as epifauna. *Modiolus undulata* and barnacles settled in large numbers on cultch and poles employed in collecting oyster spat in the neighbourhood of this bed also.

C. III Oyster Bed

This is a small bed with a limited number of eleven patches and formations located further up in the estuary along its northern bank 2.94 km from the mouth of estuary, at a distance of 19 m to 36 m from the bank Patches 1 to 9 of this bed were at a distance of 19 m from the left bank while formations reached a height of 0.4 m to 0.6 m. The area of the oyster formations varied from 6.25 sq. m. (patch 8) to 201 sq. m. (formation 10). The estimated total area of the bed was 562.22 sq. m (Table 2).

The density of oysters varied from 96 per sq. m. in formation 10 to 238 per sq. m. in patch 1. The average density of oysters and standard deviations of the same in the three beds in the estuary were found to be 198±113 in bed I, 215±90 in bed II and 158±35 in bed III. These figures indicate that there are no well defined differences in the average density of oysters in the three beds. The number of oysters per patch varied from 775 in patch 8 to 19,296 in formation 10 with an average of 7,083 oysters. It has been estimated that a total of 77,923 oysters were present in this bed (Table 1). The estimated total biomass of oysters ranged from 151.66 kg in patch 8 to 3,775.22 kg in formation 10 with an average of 1,386.31 kg for formation and the estimated total biomass of oysters in the entire bed was 15,249.48 kg. The total meat weight of oysters per patch varied from 5.73 kg in patch 8 to 142.79 kg in formation 10 with an average of 52.41 kg and the estimated total meat weight of oysters in the bed was 576.59 kg.

Biological aspects

Oysters were of the size range from 49 mm to 212 mm with an average of 103 mm for 294 oysters. 84.1% of the oysters were in the size range of 61-140 mm. Oysters below 50 mm formed only 1.7%. The weight of oysters varied between 22.5 gm and 501.5 gm. The shells of most of the oysters in these beds were massive and hence the increased weight of the oysters. The averge weight of the oysters was found to be 195.7 gm. The meat of the oysters showed a range of 2.0 gm to 12.3 gm with an average value of 7.3 gm. Males and females as well as hermaphrodite oysters were recorded. All maturity stages *viz.*, immature, maturing, ripe, partially spawned and spent ones in indeterminate phase were recorded. As in the earlier two beds, the meat of ripe oysters was cream coloured and thick and those of partially spawned and spent ones thin and watery. There was setting of only stray numbers of spat on cultch kept near this oyster bed.

Ecological conditions

There was not much difference in the epifauna and epiflora communities from those of beds I and II except that *Modiolus undulata* were found were found in comparatively smaller numbers in bed III.

Seasonal Variations in Meat of Oysters

Meat Weight

The meat weight of male oysters varied from 4.5862 gm to 17.7010 gm and that of female oysters from 5.9536 gm to 19.2248 gm (Table 3). The meat weight of oysters exhibited fluctuations in the different months (Table 4). Well-defined differences in the meat weight were not seen in relation to sex and stages of maturity. The average meat weight showed a slight decrease from 9.1469 gm in August to 8.1961 gm in September, and an increase to 11.3333 gm in November, after which it fell again. The average meat weight increased later in May to 13.3434 gm. The minimum meat weight and maximum meat weight were observed in October and November, 1975, respectively.

Percentage edibility

Like meat weight the percentage edibility also exhibited large fluctuations in all months of the year. It varied from 2.07 to 6.16 in males and from 2.10 to 7.04 in females. The percentages edibility of male and female oysters in different maturity stages showed similar ranges. The average percentage edibility increased from 3.23 in August to 4.66 in November, and decreased to 3.50 in December. After a four months period of more or less same values, it rose gradually during March-April. The average percentage edibility was 4.63 in May and decreased to 4.39 and 4.16 in May and June. The lowest value of 0.85 was seen in August and the highest of 7.04 in November.

Water

Water is the major constituent of oyster meat forming 66.48% to 89.64% wet wt. In males the content ranged from 67.59% to 89.15% wet weight and in females from 67.5% wet wt to 89.64 % wet wt. In hermaphrodite oysters it varied from 75.22% to 88.22% wet wt. The mean values of water content of oysters of the two sexes in various maturity stages did not show significant differences. The average water content decreased from 79.17% in August to 77.59% in October, increased to 83.26% in November and showed a gradual fall in succeeding months attaining 69.96% in July.

Total solids

The total solids content ranged from a minimum of 10.36% to 33.52% wet wt. Males showed variation from 10.85% to 33.52% wet wt. and females showed a similar range of 10.36% to 32.46% wet wt. In hermaphrodite oysters total solids also did not show significant difference in relation to sex and stages of maturity. The average total solids content showed an inverse relation to water content (Fig. 3). It increased from 20.83% wet wt in August to 22.43% wet wt. in September, declined to 16.73% in November and thereafter it slowly increased in succeeding months with values of 19.81% wet wt. in January, 22.65% in March and 30.03% in July.

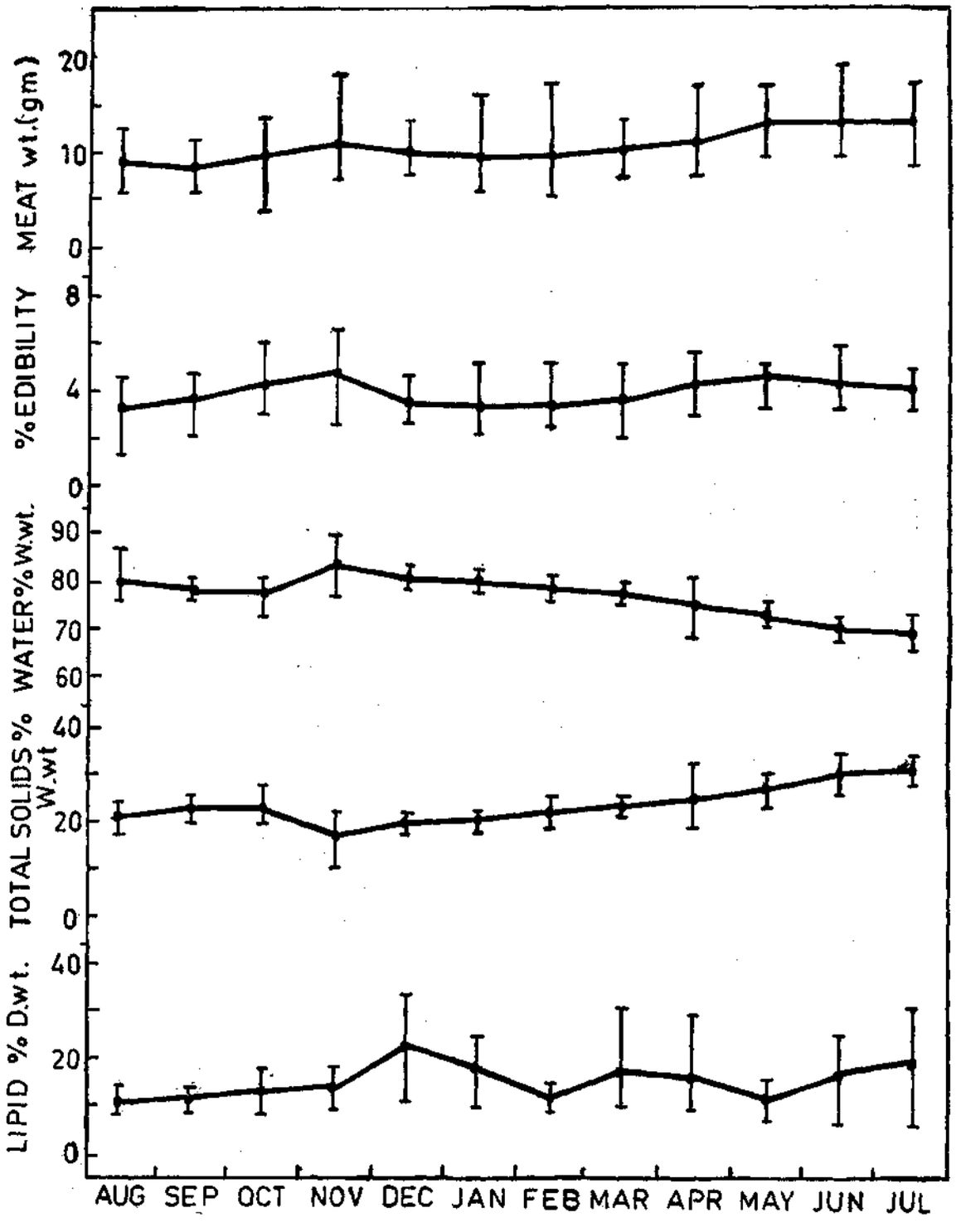

Fig. 3. Seasonal variations in meat weight, percentage edibility and water, total solids and lipid contents of *Crassostrea madrasensis* of Athankarai Estuary.

TABLE 3. *Meat weight, percentage edibility, total solids and lipid content of* Crassostrea madrasensis *of Athankarai Estuary in different maturity stages (Mean values with S.D. and ranges)*

	Meat weight (gm)	Percentage edibility	Water (% wet weight)	Totalfisolids (% wet weight)	Lipid (% dry weight)
1. Immature males ..	13.7920 ± 4.7297 (7.1370 — 17.7010)	40.40 ± 1.24 (2.65 — 5.33)	74.41 ± 3.89 (69.02 — 78.07)	25.58 ± 3.89 (21.93 — 30.98)	12.6 ± 2.7 (9.6 — 16.2)
2. Maturing males ..	12.2217 ± 2.8611 (6.8073 — 16.6650)	4.25 ± 0.72 (3.17 — 5.19)	75.67 ± 2.86 (79.25 — 78.85)	24.32 ± 2.86 (21.15 — 29.75)	13.1 ± 3.4 (9.0 — 19. 4)
3. Mature males ..	9.5747 ± 2.9836 (4.5862 — 17.3812)	3.84 ± 0.97 (2.07 — 6.09)	77.58 ± 2.37 (72.57 — 82.01)	22.41 ± 2.37 (17.99 — 27.43)	13.84 ± 6.18 (8.0 — 32.2)
4. Partly spawned males	9.0164 ± 3.1543 (6.5280 — 15.1434)	3.69 ± 1.29 (2.35 — 6.16)	80.88 ± 4.22 (77.86 — 89.15)	19.11 ± 4.22 (10.85 — 22.14)	11.4 ± 1.2 (11.0 — 13.4)
5. Spent males ..	12.0856 ± 2.2894 (9.6930 — 15.0728)	4.79 ± 1.03 (3.53 — 6.06)	75.97 ± 5.78 (67.59 — 82.13)	24.03 ± 5.78 (17.87 — 32.41)	20.6 ± 5.9 (10.4 — 28.8)
6. Recovering males ..	14.2317 ± 3.7955 (12.1834 — 16.9396)	4.83 ± 0.86 (3.49 — 5.69)	72.06 ± 3.19 (66.48 — 76.99)	27.93 ± 3.19 (23.01 — 33.52)	14.1 ± 3.4 (10.2 — 20.0)
7. Immature females ..	8.3675 ± 1.2849 (6.6042 — 10.1624)	3.06 ± 0.38 (2.44 — 3.52)	78.10 ± 0.28 (77.62 — 78.38)	21.89 ± 0.28 (21.62 — 22.38)	11.1 ± 1.4 (9.2 — 13.0)
8. Maturing females ..	8.8468 ± 1.7277 (7.0780 — 11.6386)	3.30 ± 0.74 (2.10 — 4.61)	75.95 ± 2.41 (71.73 — 78.72)	24.04 ± 2.41 (21.28 — 28.27)	14.1 ± 7.0 10.0 — 26.2)
9. Mature females ..	9.6268 ± 2.6208 (5.9536 — 14.1332)	3.68 ± 0.59 (2.30 — 4.63)	77.12 ± 2.51 (71.77 — 80.46)	22.87 ± 2.51 (19.54 — 28.23)	13.2 ± 6.3 (5.6 — 33.0)
10. Partly spawned females ..	12.9772 ± 3.7699 (7.2136 — 19.2248)	4.51 ± 1.34 (3.09 — 7.04)	79.94 ± 7.28 (67.54 — 89.64)	20.06 ± 7.28 (10.36 — 32.46)	18.0 ± 4.5 (9.4 — 32.0)
11. Spent females ..	8.2478 ± 1.8441 (6.1784 — 10.9808)	3.05 ± 0.51 (2.47 — 3.78)	77.27 ± 3.96 (71.12 — 81.45)	22.72 ± 3.96 (18.55 — 28.88)	12.3 ± 3.5 (6.2 — 23.6)
12. Recovering females ..	11.5214 ± 2.4306 (8.4414 — 15.3546)	4.03 ± 0.63 (3.28 — 4.76)	74.68 ± 3.15 (70.40 — 79.69)	25.32 ± 3.15 (20.31 — 29.60)	15.4 ± 8.1 (8.2 — 31.0)
13. Indeterminate oysters ..	11.2975 ± 3.0252 (5.8376 — 19.7056)(	3.79 ± 0.95 0.85 — 5.32)	76.46 ± 5.14 (68.62 — 86.24)	23.53 ± 5.14 (13.76 — 31.38)	15.1 ± 7.0 (6.4 — 30.8)
14. Hermaphrodite oysters (male to female sex) ..	4.0152	5.67	77.85	22.15	13.0
15. Hermaphrodite oysters (female to male sex) ..	9.0652 ± 2.6814 (5.9288 — 14.5380)	3.59 ± 1.33 (2.29 — 6.11)	82.03 ± 4.71 (75.22 — 88.22)	17.96 ± 4.71 (11.78 — 24.78)	12.6 ± 2.8 (10.0 — 18.6)

TABLE 4. *Seasonal changes in meat weight, percentage edibility, water, total solids and lipid content of* Crassostrea madrasensis of *Athankarai Estuary*
(Mean values with S. D. and ranges)

	Meat weight (gm)	Percentage edibility	Water (% wet weight)	Total solids (% wet weight)	Lipid (% dry weight)
August ..	9.1469 ± 1.9797 (5.8376 — 12.5326)	3.23 ± 1.02 (0.85 — 4.42)	79.17 ± 3.50 (76.60 — 86.24)	20.83 ± 3.50 (13.76 — 23.40)	10.2 ± 0.3 (8.0 — 12.4)
September ..	8.1961 ± 1.7936 (4.5862 — 11.3006)	3.61 ± 0.71 (2.07 — 4.73)	77.56 ± 1.50 (74.94 — 80.86)	22.43 ± 1.50 (19.14 — 25.06)	10.2 ± 0.4 (8.6 — 13.4)
October ..	9.4139 ± 2.7027 (4.0152 — 13.8000)	4.27 ± 0.90 (3.10 — 6.09)	77.59 ± 2.14 (72.57 — 80.46)	22.41 ± 2.14 (19.54 — 27.43)	12.3 ± 2.6 (8.0 — 17.4)
November ..	11.3333 ± 3.8803 (7.4966 — 19.2248)	4.66 ± 1.07 (2.70 ± 7.04)	83.26 ± 5.26 (76.74 — 89.64)	16.73 ± 5.26 (10.36 — 23.26)	13.0 ± 2.4 (10.0 ± 18.6)
December ..	9.8915 ± 2.4864 (7.2136 — 13.4362)	3.50 ± 0.51 (2.72 — 4.62)	80.17 ± 0.99 (78.99 — 82.01)	19.81 ± 0.98 (17.99 — 21.01)	22.1 ± 7.1 (11.0 — 33.0)
January ..	9.2399 ± 3.2755 (5.9536 — 15.7284)	3.32 ± 0.98 (2.18 — 5.36)	80.18 ± 1.40 (77.86 — 82.13)	19.81 ± 1.40 (17.87 — 22.14)	17.2 ± 5.9 (10.0 — 25.0)
February ..	9.2008 ± 3.1111 (5.9288 — 16.5382)	3.31 ± 0.83 (2.29 — 5.24)	78.23 ± 1.27 (76.15 — 80.41)	21.76 ± 1.27 (19.59 — 23.85)	10.9 ± 0.4 (9.2 — 13.0)
March ..	10.3985 ± 2.1350 (7.0780 — 13.4714)	3.72 ± 0.86 (2.10 — 5.19)	77.34 ± 1.20 (75.22 — 78.85)	22.65 ± 1.20 (21.15 — 24.78)	16.3 ± 7.1 (9.6 — 30.8)
April ..	10.8696 ± 3.1607 (7.7270 — 16.9396)	4.32 ± 0.86 (3.27 — 5.69)	75.64 ± 4.38 (67.59 — 81.38)	24.35 ± 4.38 (18.62 — 32.41)	15.1 ± 5.8 (9.4 — 28.8)
May ..	13.3434 ± 2.2751 (9.6182 — 17.3812)	4.63 ± 0.70 (2.93 — 5.54)	73.32 ± 1.38 (73.91 — 76.48)	26.67 ± 1.38 (23.52 — 28.09)	10.9 ± 2.7 (6.4 — 16.0)
June ..	13.6450 ± 2.9631 (10.5742 — 19.7056)	4.39 ± 0.91 (3.28 — 6.06)	70.27 ± 1.53 (68.62 — 73.63)	29.72 ± 1.53 (26.37 — 31.38)	16.2 ± 5.8 (6.2 — 26.2)
July ..	13.2118 ± 2.6235 (8.4414 — 17.7010)	4.16 ± 0.66 (3.28 — 5.33)	69.96 ± 1.74 (66.48 — 71.89)	30.06 ± 1.74 (28.11 — 33.52)	19.0 ± 7.7 (5.6 — 32.0)

Lipid

The lipid content showed a range of 5.6% to 33.6% dry weight. In males the lipid content varied from 8% to 32.2% and in female oysters from 5.6% to 33% dry wt (Table 3). The content of hermaphrodite oysters showed a lower range of 10% to 18.6% dry wt. As in the case of water and total solids, the lipid content of male and female oysters too did not show significant difference correlated with maturity stages and there is overlapping of the lipid content of oysters in different stages of maturity. While the lipid content of individual oysters showed wide fluctuations in various months (Fig. 4) the average lipid content increased from 10.2% in August to 13.0% in November and 22.1% in December. Thereafter, it decreased to 17.2% in January and 10.9% in February. It increased again to 16.3% in March, decreased to 10.9% in May and rose to 16.2% in June and 19.0% in July. Thus average lipid content showed a rise thrice in an annual period in December, March and June-July.

Discussion

The total area of the three oyster beds in the estuary is 15,621.91 sq. m or 1.56 hectares. Of this, the bed II alone has 13,692.85 sq. m. an area constituting 87.6% of the total oyster bed area. The area of the bed I is only 1366.84 sq. m. which is 8.7% of the total area while the III bed has an extent of 562.22 sq. m forming 3.7% in the total area. In the I bed the area of a single oyster patch ranges from a very small size of 0.25 sq. m. to 176.5 sq. m. The oyster patches and formations in III bed are slightly bigger with a range of 6.25 sq. m. to 201 sq. m. In the II bed thirty patches are 3.25 sq. m. to 85.37 sq. m. in area while eighteen formations 1, 3, 4, 5, 7, 11, 12, 15, 20, 22, 25, 30, 32, 35 to 37, 45 and 48 are larger with an area of 113.12 to 621.62 sq. m. Oyster formations 6, 8 and 23 are the largest beds in the estuary with areas ranging from 2,207.37 sq. m. to 2,705.62 sq. m.

The density of oysters in the three beds is highly variable with a range of 43 to 520 oysters per sq. m. The highest densities of 300 to 480 or 520 per sq. m. have been recorded in patches 19, 29 to 32 and 35 in I bed and in formations 1 to 5 in II bed.

There are a total of 34,72,572 oysters in Vaigai estuary at Athankarai including 3,44,116 oysters in I bed, 30,50,533 in II bed and 77,923 in III bed. The largest number of oysters are present in the II bed which also has the most extensive area of oyster beds. The richness of the density and abundance of oysters in the II bed is ascribed to the constant mixing when the estuary is connected with Palk Bay of fresh water coming from the upper portion of the estuary and sea water from seaward side during the course of tidal oscillations every day. The constant mixing of fresh and sea water appears to be very favourable for the growth and increase in population of *Crassostrea madrasensis* in the estuary. Rao and Nayar (1956) have pointed out that growth of *C. madrasensis* was better in Adyar estuary than in marine environment of Madras Harbour.

The estimated total biomass of oysters in Vaigai estuary is 3,88,921.29 kg and the estimated total meat weight of the oysters 13,674.10 kg. The estimated total biomass of oysters in bed II is largest being 3, 25, 164.92 kg and the total meat weight of oysters in the bed is 11,111.72 kg. In bed I the the estimated total oyster biomass is smaller, 48. 506. 89 kg and the total meat weight is 1,985.79 kg corresponding to the much smaller total area of bed. In bed III where the total area is only 562.22 sq. m. the total oyster biommass is 15,249.48 kg and the total meat weight 576.59 kg.

The size, total weight and meat weight of oysters in the I and II beds do not show marked differences. The total weight and meat weight of oysters in bed III are higher than in the other two. The average total weight and meat weight of oysters in bed III are 195.7 gm and 7.3 gm respectively compared to 112.6 gm and 4.7 gm in bed I and 106.5 gm and 4.1 gm in bed II.

Although seaweeds and epifauna and pests belonging to different groups such as sponges, whelks, weaving mussels, polychaetes, amphipods and barnacles are present on oysters, only *Modiolus undulata* and barnacles seem to be serious pests. *Modiolus undulata* are competitors for space as they settle in large numbers on the surface of live oysters and over empty oyster shells where oyster spat could set and grow as was noticed in bed II. The barnacle *Balanus amphitrite communis* also settles in large numbers on cultch in the lower and middle portions of the estuary. The danger posed by the boring polychaetes of *Polydora* sp. does not appear to be much as they are not common in the area.

The pea crab *Pinnotheres* sp. has been reported in *Ostrea* (=*Saccostrea*) *cucullata* by Awati and Rai (1931) and in *C. gryphoides* by Durve (1964) along Bombay coast but pea crabs have not been recorded in *C. madrasensis* in Athankarai estuary. Algae, sponges, hydroids, polychaetes and barnacles have also been recorded on the surface of valves of *C. cucullata* in marine environment by Awati and Rai (1931). Preda-

tory whelks and hermit crabs are not common in Athankarai estuary. In U.S.A. the oyster whelks *Urosalpinx cinerea, Eupleura caudata* and *Thais* spp. are destructive to the American oyster *Crassostrea virginica* (Galtsoff, 1964).

The investigation has brought to light considerable population of *Crassostrea madrasensis* in Athankarai estuary. Until now the valuable oyster resources are not being utilized properly by the people of the area. The oysters of the estuary are nutritious and as they occur in very large quantities in beds, the natural stocks should be exploited for consumption at least to some extent. Oyster shells could be burnt and converted into lime. The need for utilization of natural stocks is all the more great as otherwise there is heavy mortality of oysters in certain years due to steep increase in salinity following evaporation of water or prolonged flooding of estuary with freshwater due to heavy rains.

The existence of the large population including spawners and good spatfall which takes place in the estuary are favourable for culturing *C. madrasensis* in the estuary. The estuary is connected with the sea for a greater part of the year and phytoplankton is available for oyster larvae, spat and adults. Oyster spat could be collected in large numbers using oyster shell, concrete piece and tile cultch and growing oysters could be reared to marketable size in trays kept on racks. (Rao 1976, Rao, Sivalingam and Unnithan, 1983). Spatfall took place from January to April in Athankarai estuary and generally only small numbers of spat were seen to set on cultch in the other periods of the year. Studies on the growth of spat and oysterlings indicate that growth of oysters is rapid in the estuary and an average size of 87 mm. and maximum size of 110 mm are attained at the end of one year. Average and maximum sizes of 90 mm and 130 mm are attained at the end of 14 months and thereafter, there is retardation of growth. The fast rate of growth will be very advantageous for conducting oyster culture successfully in the estuary.

In some years there is large increase in salinity in the premonsoon months and in certain other years there is prolonged flooding of the estuary with freshwater in the north-east monsoon season causing large mortality of oysters. In October, 1977 following very heavy monsoon rains, the Vaigai Estuary at Athankarai was in spate and as a consequence of continuous flow of freshwater there was a large scale mortality of oysters in the natural beds in the estuary. However, there was re-colonization of oysters and revival of oyster beds in 1979. Large-scale mortality of oysters due to environmental factors has been mentioned by Yonge (1960) and Galtsoff (1964) in *Ostrea edulis* and *Crassostrea virginica* in which freezing temperatures in winter, shooting up of temperature in summer or flooding of estuaries lead to extensive mortality of oyster beds. During such times cultured oysters have to be transferred to racks installed in nearby coastal waters. Extremes of salinities are prevalent only in some years in Athankarai estuary and is not a regular feature. Commercial exploitation of oysters from natural beds will lead to depletion of resources. Therefore, when a beginning is made in the utilization of resources, it is very desirable that culture practices are adopted.

The average meat weight of *Crassostrea madrasensis* shows a slight increase in November followed by a fall and rise in April-July period. The average percentage edibility which is an index of the condition of meat shows a similar trend and is higher twice in the annual period in November and again in April-July. In *Crassostrea madrasensis* of Ennore backwaters Venkataraman and Chari (1951) found the percentage edibility to be low in July and fairly high in October and Rao (1956) observed three peaks in meat weight and percentage of meat weight in whole weight of oysters in February, July-August and November.

The maximum water content of *C. madrasensis* of Athankarai area 89.64% wet wt. is higher than 85.04% wet wt. recorded in the same species of Ennore backwaters by Venkataraman and Chari (1951) and much higher than 74.5% wet wt. recorded by Giese (1969) in the mussel *Mytilus edulis*. Total solids constituted on an average 16.73% to 22.65% wet wt. in the period August to March in *C. madrasensis* of Athankarai Estuary but steadily increased to 24.35% wet wt. in April and as much 30.03% wet wt. in July.

Venkataraman and Chari (1951) found that the maximum lipid content of *C. madrasensis* occurring in Ennore backwaters was 14.8% of dry wt. The maximum lipid content in the present studies has been observed to be more than double that value, being 33% dry wt. The Athankarai oysters are much richer in lipid than Ennore oysters. Compared with the lipid values given by Galtsoff (1964) for the American oyster *Crassostrea virginica* (12.8% of dry wt.) and the European oyster *Ostrea edulis* (13.2%) recorded by Gaarder and Alsaker (1941) the lipid content of oysters recorded in this work (33% dry wt.) is much higher. The present studies indicate inverse relationship between water and lipid contents of oysters in the estuary. In this context, it may be mentioned here that similar findings have been made by Masumoto, Masumoto and Hibino in

Ostrea (=*Crassostrea*) *gigas* and by Durve and Bal (1961) in *Crassostrea gryphoides*.

Absence of well-defined quantitative changes in meat weight and chemical constituents in *C. madrasensis* of Athankarai Estuary in relation to reproductive stages is attributed to the absence of periodicity in breeding of the species in the Athankarai estuary resulting in constant acumulation of organic constituents in the body of oysters for maturation of goands and energy requirements of the body.

One interesting finding of the present work is that the levels of water, total solids and lipid of meat of male and female oysters of Athankarai estuary are more or less same and there are no distinct differences in relation to sex of the oysters. However, the lipid content of hermaphrodite oysters changing from male to female phase, is lesser than that of males or females. This could be ascribed to breakdown of lipids during the course of disintegration of gonads of one sex for the formation of reproductive follicles of the other sex.

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5

BIOLOGICAL ASPECTS OF OYSTERS

M. E. RAJAPANDIAN[1] AND C. T. RAJAN[1]

INTRODUCTION

Purchon (1968) has stated that about thousand articles are being published annually on molluscs and out of these 300 to 400 articles deal with oysters. This is mainly because oysters enjoy a wide range of distribution and are ubiquitously present in different habitats. Among marine organisms the oysters are one of the most widely cultured organisms. These facts account for the voluminous literature on oyster biology that have come out during the past fifty years.

Considering the current interest in our country in developing oyster culture as an industry this chapter aims to place all available information on oyster biology to make it useful to scientists involved in oyster studies.

HABITAT AND DISTRIBUTION

Along the Indian coasts the occurrence of oysters of only two genera *viz.*, *Crassostrea* and *Saccostrea* has been reported. Of these *Crassostrea* spp. are widely distributed along both coasts, while *Saccostrea* represented by *S. cucullata* is comparatively not so abundant. Of the eight species listed by Awati and Rai (1931) the three species *C. madrasensis*, *C. gryphoides* and *C. rivularis* are important as resources. The other species are of little or of no consequence. These species thrive well in coastal and estuarine conditions. But *Crassostrea madrasensis* is widely distributed along both coasts, whereas *C. gryphoides* and *C. rivularis* are restricted to only North western coastal zones. Naturally even the very little information we have on the Indian oyster species is more on *C. madrasensis* as reflected in the work of Hornell (1910, 1916), Paul (1942), Rao (1951, 1956, 1983), Rao and Nayar (1956), Durve and Bal (1962), Rao (1974, 1983), Nayar and Mahadevan (1983) Mahadevan (1983), Purushan *et al.* (1983), Reuben *et al.* (1983), Joseph and Joseph (1983), Thangavelu and Sundaram (1983), Thangavelu and Muthiah (1983), Rajapandian and Rajan (1983) and Samuel (1983).

Descriptions of the external morphology of the oyster shells and the internal anatomy of the oysters are well documented by Galtsoff (1964), Awati and Rai (1931) and Purchon (1968). Korringa (1952) has also made an exhaustive review of the description and functional differentiation of different body systems of the oysters. Hence further description of these in respect of *C. madrasensis* will be redundant in this chapter as there are very few differences in the above aspects described.

The biology of oysters is greatly influenced by the environmental factors in the habitat. Feeding, growth maturation, spawning, development and setting of oyster spat are greatly influenced by the varying environmental factors such as temperature, salinity, pH, dissolved oxygen, sediments and food. By far temperature and salinity of the water greatly influence the growth, survival, reproduction and larval growth and metamorphosis.

Increased industrial activity has led to increased release of toxic substances including hevy metals into the environment. This material may be discharged directly into estuarine and marine environments or may accumulate there through water run off. Many of these substances such as arsenic, cadmium, chromium, copper, lead, mercury etc. can be concentrated by oysters and thus become a potential hazard to human beings who consume them. Further these materials may exert a lethal or sublethal effect on different stages

[1] *Present Address :* CMFRI, Research Centre, Tuticorin-628 001.

of the life-cycle of oysters, with consequent influence on population abundance. Hence monitoring of heavy metals has to be carried out in order to find out any unusual increase in heavy metal content before it becomes a health hazard.

Food and Feeding

Food

The food of oysters mainly consists of organic detritus and phytoplanktonic organisms such as diatoms and nannoplankters. The widely accepted view on food of oysters is that phytoplankton constitutes the principal source of shellfish food. Spores and particulate matter of seaweeds are also found in the stomach. Nelson (1947) found that the abundance of the diatom *Skeletonema* has been responsible for the healthy growth and good condition of oysters of Delaware Bay. Jorgensen (1975) observed correlation between rapid growth of oysters and the presence of the diatoms, *Skeletonema* sp. *Chaetoceros* sp. and *Thalassiosira* sp. Epifano (1979) comparing nutritional effects of different algal species, found a combination algal diet with *Isochrysis galbana* and *Thalassiosira pseudonana* promotes better growth than the diet consisting of only a single species.

These microorganisms and other particulate matter are filtered from water by ciliary action of gills and transported to the mouth and from there passed to the stomach and digestive diverticula for digestion and absorption. The mechanism of feeding in adult oysters has been well documented (Nelson 1938, 1960, Korringa 1952, Menzel 1955, Jorgenson 1966, 1975, 1976, Owen 1974). Galtsoff (1964) provides an excellent account of the anatomy and physiology of gills.

Process of ingestion

The food particles carried by streams of water pass through the gills and become entrapped, bound in mucus and are transferred towards food collecting furrows. Masses of collected food are converged in strings of mucus to the labial palps where particulate matter is sorted either to be passed on to the mouth or rejected as pseudofaeces. The labial palps on either side of the mouth play an important role in sorting the food. The outer palps join together above the mouth, where they form the upper lip and the inner palps fuse together below the mouth as lower lip. A narrow cavity found between the inner palps is known as median gutter and the spaces between the external and internal palps are termed lateral gutters. These are the principal paths by which the food is converged to the mouth. Bernard (1974) suggests that the ciliated ridges of these grooves (gutters) are mucus-reducers with ability to reject the entire mucus-particle load on their surface if the size of the particles is large. Smaller particles with lesser amount of mucus follow a deeper sinuous path of ciliary tracts towards the mouth. The final acceptance or rejection of particles is determined mainly by the amount of mucus secreted by the gills and palps.

Process of digestion

The digestion and absorption of food in oysters is effected extracellularly in the stomach and intracellularly in the digestive diverticula. Yonge (1926), George (1952) Levine (1946) and Newell (1953) by their investigations found with conclusive evidence the presence of amylase, glycongonase, oxidase and lipase in the stomach. Yonge (1926) viewed that protein and fat digestion occurred only intracellularly within the wandering phagocytes and starch is digested only extracellularly by the action of thyliamylase. In general the phagocytosis i.e. intracellular digestion and absorption mainly occur in the ducts and tubules of digestive diverticula. These cells are the only absorptive areas in the gut (Purchon, 1968). Further phagocytes play an important role in the removal of waste materials from digestive diverticula. They discharge waste materials in the intestinal lumen by bursting off.

Extracellular digestion in the stomach is effected by the mechanical turning of the stomach and chemical dissolution of the crystalline style. The movement of the food in the alimentary tract is mainly accomplished by the strong ciliary motion of the epithelial lining of the system.

In the stomach, the crystalline style performs a number of functions that are relevant to the digestive process (Purchon, 1968). The style rotates (Yonge, 1969) at a rate of 90 revolutions/minute. By the rotatory movement the food material is drawn to the stomach more rapidly than it could enter under the impetus of the oesophagal cilia. Further the movement of the style stirs the general contents of the stomach and brushes these against the corrugated ciliary sorting areas which may segregate nourishable particles and eject them into the midgut. As the head of the style gently rubs the contents of the stomach against the gastric shield, the food particles may undergo certain amount of trituration and partial reduction of the particle size. The style slowly dissolves liberating the amylolytic enzymes which initiate a preliminary phase of extracellular digestion in the stomach.

The digested and undigested particles flow in the digestive tubules of the digestive diverticula. Purchon (1955, 1968) suggests that the cavities of the stomach, the ducts and the tubules of the digestive diverticula are in communication and that the fluid may pass to and out of the duct as a result of changes in pressure on the system. This could be effected by local changes in the tonus of the muscle fibres which are present in the wall of the stomach and around the individual ducts and tubules of the diverticula (Purchon, 1955). In the larva of oyster, rudimentary diverticula having two simple sacs exhibit rhythmic alternate contraction to bring about the movement of the particles in alimentary tract. The contraction of diverticula is effected by a slender shield of muscle fibres that pass over the top of the diverticula (Awati and Rai 1931, Purchon 1968).

From the midgut the rejected materials by the typhlosole channel reach the hindgut wherefrom these materials are further thrust by epithelial cilia to the rectum and anus.

The oyster is also capable of absorbing dissolved organic matter in the water through the surfaces of the gills, palps and mantle. Owen (1974) says that the gills of bivalves possess active carrier-mediated transport systems for the absorption of neutral amino acids and hexose monosaccharides. Further Pasteels (1967) observes that amoebocytes within the gill may serve to translocate such materials. Vitamins such as riboflavin, calcium pantothenate, thiamin and pryidoxine when added to the medium have significantly increased the rate of growth of *O. virginica* and *Ostrea lurida* (Davis and Chanley 1956). Pomeroy (1952) and Pomeroy and Haskin (1954) have concluded that significant amounts of phosphate and calcium ions are derived from the water, thus partially fulfilling the requirements of oysters for these ions both for carbohydrate metabolism and shell deposition. According to Jorgensen (1955) 0.05 mg/litre amino acid is available in sea water. Jeffrey (1966) has observed that potentially useful classes of lipids are available in the sea water in the range of 0.5 to 0.6 mg/litre. Lewis and Rakestraw (1955) have reported carbohydrates in coastal lagoons present at 8 mg/litre. In view of the potential absorptive powers, the oysters can assimilate considerable quantity of soluble organic substances from the sea.

Factors affecting the feeding of oysters

Loosanoff and Nomejko (1946) have recorded the pumping rate of oysters as 34.0 l./hour at 25°C. At temperature of 28°C to 32°C a maximum pumping rate of 37 l. to 40 l./hour was observed for 5 to 15 minutes period. According to them the optimum pumping rate occurs at temperature of 25°C to 30°C. Although higher temperature regime increases the pumping rate for a short while after certain time the pumping rate slowly decreases.

Loosanoff and Engle (1947) have reported that the rate of pumping is influenced by the density of microorganisms. In some cases certain inhibiting substances such as metabolites of microorganisms influence the pumping rate. In such instances the rate of pumping is reduced. Under high concentration of microorganisms, the production of pseudofaeces increases. A reverse relation is observed in the production of true faeces.

Theede (1963), Davids (1964) and Thompson and Bayne (1972) have demonstrated that the particle concentration affects filtration rate. In *O. edulis* and *C. angulata* the secretion of mucus increases with higher particle concentration and the ctenidia are blocked.

Growth

The growth of oyster is expressed in terms of increament in length (height of the shell) and weight. The shell growth is correlated with growth of living tissues.

(a) C. madrasensis

Hornell (1910), Paul (1942) Rao and Nayar (1956), Rao (1974), Nagappan Nayar and Mahadevan (1983), Rao *et al.* (1983), Purushan *et al.* (1983), Joseph and Joseph (1983) and Reuben *et al.* (1983) have made investigations on the growth of *C. madrasensis.* It is observed that the growth of spat is rapid during the first 3 months. A size of 38 mm is attained in 90 days registering a growth of 12.6 mm per month and at the the end of first year oysters attain an average size of 84 mm with a total shell and meat weight of 120-130 g per oyster (Nagappan Nayar and Mahadevan, 1983). Rao *et al.* (1983) have observed that an average size of 86.7 mm and maximum of 110 mm were reached at the end of one year at Athankarai. Purushan *et al.* (1983) recorded an average growth of 60-62 mm in a period of 5-5½ months in Cochin backwaters. Joseph and Joseph (1983) found that oysters of Mulki estuary grew to the marketable size of 70 mm in 7 months. Reuben *et al.* (1983) reported a growth of 77-81.8 mm in 12 months in the oysters of Bheemunipatnam backwaters.

(b) C. gryphoides

Durve and Bal (1962) recorded a growth of 37.2 mm and 47.9 mm at the end of six months and one year

respectively in *C. gryphoides* of Kelwa backwaters. The growth of this species is thus distinctly slower than that of *C. madrasensis*.

(*c*) *C. rivularis*

No information is available on the growth of this species.

Growth of oysters is largely influenced by the availability of food and hydrographic conditions. The growth and survival of oyster populations along the east coast of India appear to be relatively better perhaps due to the stable salinity and temperature conditions prevailing along the coast.

In temperate waters growth of oysters is almost restricted to summer months. Loosanoff and Nomejko (1949) observed that in *C. virginica* there is no increase in size, volume or weight during winter months (December-March) while during the eight months, April to November, most rapid growth in length occurred. Quayle (1950) and Cahn (1950) stated that *C. gigas* would require two full summer seasons to attain marketable size of 75-85 mm.

Conditions of Oysters

Condition of oyster is recognised as the degree of fatness of an oyster or the extent to which the meat fills the shell cavity. The oyster shell grows to accommodate the soft body. However, the body size of oyster undergoes changes and such changes are associated with the breeding cycle. This is accomplished by development of an increase in size of the reproductive organs followed by a considerable reduction in size after spawning. This process is followed by a slow increase in body size. In temperate waters increased level of glycogen has been associated with this phase. But in tropical waters this phase of glycogen accumulation has not been observed. Changes in the meat content of an oyster are important to the culturist for these greatly affect the meat yield and financial returns. Thus knowledge of the seasonal fatness cycle is most important for successful marketing.

The condition factor is measured as a ratio comparing the dry meat weight (oven dried at 90-100°C) of the oysters to the volume of the shell cavity.

$$\text{Condition factor} = \frac{\text{Weight of dry meat} \times 1000}{\text{Volume of shell cavity}}$$

In *C. madrasensis* the condition may be high if the value is above 140. Values below 70 indicate the poor condition of oysters. It has been observed that along Tuticorin bay the condition factor reaches high values during February-March.

Reproduction

Reproduction of oysters in contrast to many other animals is simple. The gonad of oysters is located under the mantle and consists of branching tubes and follicles on each side of the body encasing the visceral organs. The follicles containing germ cells and follicular tubules with a well developed epithelium to facilitate the passage of gametes. As the gametogenetic activity of oyster increases with accummulation of mature ova and sperms the gonad becomes thick. The ripe eggs and sperms pass along a series of tubes by ciliary action in the tubes which finally merge in a tube along the dorsal side of the body. Two separate systems of genital canals are found one on each side of the oyster which open into the suprabranchial chamber and from there discharged to the exterior.

Maturation of Gonads

Paul (1942) and Rao (1951) have recorded partially spent conditions of the gonad of *C. madrasensis* at Madras all round the year. Among oysters found in Adyar backwaters of Madras and in Tuticorin Bay (Gulf of Mannar) gonads with fully ripe and partially spent conditions have been recorded in high percentages during February-April and July-September. Nayar (1977) and Rajapandian and Rajan (1983) have observed that in the Gulf of Mannar, although the spatfall was observed throughout the year, the peak of gametogenetic activities were recorded during the months February-March (64-78%) and August-September (59-73%). Further they have observed that the diurnal variations in minimum and maximum temperature have well defined relation to the development of gonads and spawning of oysters. The highest value observed during February initiates the maturation of oysters and spawning continues till May. The drop in temperature during the months of June and July showed corresponding decline in the spawning activity of the oysters. In September this value of temperature increased and there was rise in the spawning population.

Cole (1942), Bargeton (1942) and Loosanoff (1942) have described the process of maturation of gonads and the factors influencing them. Among exogenic factors temperature is much more important than any other factors. Rand (1973) in examining the breeding habits of marine animals in time and space concluded

that in Northern climate the animals are characterised by single synchronous spawning in a year, temperate climate by two spawnings and tropical climate by year round spawning. In northern climates gametogenesis sets in European oysters during the middle of May and spawning commences in early June and continues till July or early August.

Obviously, sometimes it is difficult to separate the effects of temperature at different latitudes. Loosanoff and Engle (1942), Korringa (1957) and Galtsoff (1964) have given evidences for the existence of different physiological races of oysters, each with its own threshold spawning temperature. In *C. virginica* there are races that spawn at 17, 20, and 25°C in northern, central and southern latitudes respectively (Stauber, 1950).

Sex ratio

Oysters generally are dioecious but hermaphrodites are not uncommon. It has been well documented that young oysters function primarily as males (60-70%) and later become female. In oysters of the 'O' year class which are up to 78 mm in size 75% are males and in one year old and above, ranging in size from 80-118.5 mm females represent 72% (Nayar *et al.*, MS).

Hermaphroditism

True and functional hermaphrodites probably do not occur but are only transitional phases containing both sperms and developing eggs. Galtsoff (1964) has observed hermaphrodites only among 2, 3 and 4 year old oysters of *C. virginica* and none in 5-8 year old oysters. Rao (1953, 1956) recorded hermaprodites in *C. madrasensis* throughout the year. Further his observations on the changing pattern of sex from male to female and female to male evidently shows that hermaphroditism is a feature found during the transitional phase of sex change. From our observations on the oysters, *C. madrasensis* at Tuticorin hermaphroditism is a feature not frequently found in ripe oysters, but occur in stray instances in spent and recovering stages.

Fecundity

In the larviparous oyster, *Ostrea edulis*, the egg is about 100 μ in diameter and is very large when compared to those of the oviparous oysters *Crassostrea*. The egg of *C. madrasensis* is about 48-60 μ in diameter.

Cole (1941) found the number of eggs spawned by *O. edulis* to be 91,600 by one year old oysters and 218,100, 462,000 and 902,000 eggs were spawned by 2 3 and 4 year old oysters. Cerruti (1941) stated that the number of eggs depends on the size of oyster than the age.

Oysters of the genus *Crassostrea* release more eggs then larviparous oysters. In a season *C. virginica* can release 100 to 200 million eggs (Galtsoff, 1964). In *C. gigas* almost same numbers of eggs per spawning have been recorded (Tomiyama, 1980). During the course of our investigations in *C. madrasensis* a few ripe oysters were selected and forced to spawn by manipulation of temperature. At the onset of spawning the oysters were placed in individual glass trays of 3 l. capacity and allowed to spawn. After completion of spawning the oysters were removed from the tray and the contents were released into a beaker and made up to 10 litres. After proper stirring 100 ml sample was taken and 1 ml of 1% formalin was added to it. After further stirring 1 ml subsample was drawn on a Sedgwick Rafter chamber and the number of eggs present counted. By verifying a few more subsamples it was found that the number of eggs per spawning amounted to 10-15 millions.

Spawning

The two genera of oysters, *Ostrea* and *Crassostrea* have different spawning habits. In *Ostrea* the eggs when released from the gonad are retained in the mantle cavity while the sperms are discharged to the exterior. Eggs are fertilized by sperms from outside and the larval life partly takes place inside the shell before being released into the water. In *Crassostrea*, at spawning both eggs and sperms are discharged directly outside into the open water where fertilization and all subsequent development take place. The process by which the sperms and eggs are discharged is completely different. The sperms are discharged by the contraction of muscles in the walls of the genital ducts. The sperms carried away by the outgoing water current, appear as a dense white stream emerging from between the valves which quickly disperses in water from the exhalent side.

The spawning process is more complex in female oysters unlike the male. The female rhythmically ejects the eggs through the inhalent side. The process is controlled by the edge of mantle folds which can open and close like a zipper. The tentacles of the inner lobe of the mantle act like the components of a zipper, and keep the curtain closed except for a small opening, when spawning takes place. The discharged eggs from the ovary first reach the epibranchial chamber. At this the two edges of the mantle merge and seal the

infra and supra branchial cavities. The adductor muscle is relaxed at this point, resulting in the passage of eggs outside the gill chamber. At this stage the contraction of the adductor muscle forcefully ejects the eggs out through a narrow opening along the inhalent side. This process occurs at regular intervals.

Fertilization

In recent years studies on this aspect of biology have assumed new dimensions since innumerable workers are engaged in perfecting techniques in the artificial propagation of oysters. Investigations on induced spawning, artificial fertilization and laboratory rearing of oyster larvae have provided a fund of information on this aspect of biology.

Eggs lose their fertilizability totally at the end of 24 hours in temperate climate. In tropical conditions it is lost within 4 hours. The fertilizing power of sperms lasts for 3 to 4 hours. Temperature and dilution factors decrease the fertilizing power of sperms (Dupuy *et al.*, 1977). Concentrated suspension of sperm stored at 10-12°C will increase the longevity upto 24 hours. Therefore to ensure optimum fertilization freshly released eggs and sperms should be mixed instantaneously. A suspension of eggs from different females and sperms from several males when mixed would result in optimum fertilization and normal embryonic development. Abnormal fertilization can occur if large proportions of sperms are mixed with small quantities of eggs. This may result in several sperms penetrating the membrane of a single egg and this phenomenon, polyspermy will cause irregular cleavage of egg. Dupuy *et al.* (1977) observed that a delay of one hour or more in adding sperms to newly spawned eggs will increase the incidence of abnormality among the larvae.

Development

In *C. madrasensis* after fertilization the first polar body is observed within 20 to 40 minutes and subsequently the second polar body appears. The first cleavage occurs immediately after the formation of the two polar bodies and the cells in the animal pole divide resulting in the formation of the 8 celled stage. One of the cells (macromeres) formed in the first cleavage retain the identity and remains at the vegetal pole as the macromere. Subsequently after the 6th division, a roughly spherical morula is formed. The gastrula stage is reached between 5 and 6 hours after fertilization. At this stage the larvae start swimming upwards.

The straight hinge stage or 'D' shelled stage, the first veliger stage is reached at the end of 20 hours. The larvae actively swim and collect food with the help of the velum which is formed by an outgrowth of the lateral parts of prototroch area in two semicircular folds or lobes bearing large cilia along their margins. The margin of the velum possesses large cilia which help the larvae to swim and the small cilia present at the base of velum (aboral cilia) direct food particles to the stomodaeum. The larvae at this stage measure 60-70 μ in length (anteroposteriorly).

In larviparous oysters, *Ostrea* early embryonic development and transformation of larvae takes place in the inhalent chamber. These larvae cover densely the surface of the gills and lower mantle lobe which gives the appearance of sprinkled flour. At the time of their release the congregation of larvae are purplish black. After fertilization the larvae are retained in the gill chamber for 7 to 8 days and attain a size of 220 μ before release (Yonge, 1960).

The sequence of events in the development and growth of the larvae of the American oyster *Crassostrea virginica* and the Pacific oyster *C. gigas* are almost similar to that of *C. madrasensis* under laboratory and natural conditions. The larvae attain eyed stage on 13th day in all these oysters and setting takes place on the 14 to 15th day after fertilization. The growth of spat has been observed to be faster in *C. madrasensis* than in *C. virginica* and *C. gigas* which may be probably due to the differences in the prevailing temperature.

Variation in salinity and temperature greatly infuences the growth and settlement of larvae. Growth and setting of the larvae have been observed to be optimum when salinity and temperature regimes are steady. Davis (1958) demonstrated that lowering of salinity from normal level to 15‰ has not resulted in mortality of the larvae. However, at salinities below 12.5‰, 90-95% of the larvae died indicating that they are lethal.

Optimum growth and setting of the larvae of *C. madrasensis* have been observed at salinities 28.0 to 31.5‰ and at temperatures of 25° C to 27.0°C. Dupuy *et al.* (1977) have successfully settled the larvae within 9-11 days at salinity ranges of 15 to 20‰ at a temperature of 27°C and the larvae of *C. gigas* at 20‰ and in 27°C.

Larval food

It is known that some areas are excellent for fattening or growing oysters but not good for spat settlement

and areas where settlement is rich, offer poor conditions for the growth of oysters. Nelson (1950) suggested that the appropriate nannoplankton food for oyster larvae is not found to be useful for adults whereas diatoms and dinoflagellates might fatten adults but are of little use to larvae. Cole (1937), Loosanoff and Davis (1953), Walne (1956) and Ukeles and Sweeney (1969) have demonstrated that the best food for larvae are the phytoflagellates, belonging to the genera *Isochrysis*, *Monochrysis* (*Pavlova*), *Pyramimonas* and *Dunaliella*. The size of these phytoflagellates range from 5 to 8 μ and are highly motile. They are found in the surface and the column waters and are easily filtered by the oyster larvae. Under normal temperature and salinity conditions the straight hinge larvae of *C. madeasensis* need 4,000 to 5,000 cells per larva per day. The intensity of feeding increases with growth. At the time of settlement, 10,000 cells per larva per day is ideal (Nayar *et al.*, 1984).

Diseases and parasites

Large scale mortalities of oysters have occured from time to time for which no rational explanation could be given. Even after exhaustive investigation, the exact cauative factors responsible for such catastrophic outbreaks could not be recognised. Recent studies on histopathological aspects have thrown some light on the possible factors responsible for many instances of large scale mortalities.

Some epizootic viruses are suspected to be the agents causing such diseases. The reasons for such mortalities at Malpeque Bay and Prince Edward Island, U.S.A. during 1915 have been ascribed to viral diseases. The oyster population in these affected areas reached its former level of production after several years and it has been surmised that current populations are resistent strains that have been developed from survivors. The evidence that the infectious agent is still present is suggested by the fact that nonindigenous oysters are susceptible to the disease and die within the first or second year after the introduction (Aron Rosenyfield, 1967). Many microbial diseases are reported from all over the world. The same disease which has been observed in oysters of Japanese waters has been noticed in those in the Nasselle river, Willapay Bay, Washington.

'Dermo' a dreaded disease causing heavy mortality among eastern American oysters was first identified as fungal disease caused by the fungus *Perkinsus marinus* (= *Dermocystidium marinum*). Oysters affected by 'dermo' show watery digestive gland, thin mantle and atrophied gonads. Mycelial disease or gill disease is common in *C. gigas*. Shell disease caused by the fungus *Ostracobiabe implexa*, inflicted heavy mortality among Dutch oysters during 1930 (Korringa, 1952). This is characterized by the formation of pustules on the inner shell surface.

Minchinia nelsoni, known as MSX disease has caused heavy mortality among the oyster population in Delaware Bay, U.S.A. during 1955. *M. nelsoni* is a haplosporidian very active during summer months. Similarly the dreaded disease known as SSO caused by *M. coastalis* takes heavy mortality of oysters. *Hexamita* a commensal flagellate of oyster causes the disease known as Pit disease. Korringa (1952) has stated that in Dutch waters the 'pit disease' is caused by the prolific multiplication of *Hexamita* under low temperature conditions.

Several metazoan parasites have been found in many species of oysters. Most of these organisms are the larval stages of helminths having oysters and other bivalve molluscs as intermediate hosts. The larval trematodes of the genus *Bucephalus* have been found in American, European, Japanese and Indian waters. Although these parasites do not cause heavy mortality among oysters, they enter the gonadal tissues and cause damage resulting in the sterility of the oysters (Joseph, 1978). Samuel (1978) has described a digenic nematode parasite which is common in the gonads of *C. madrasensis* and causes sterility of the gonad.

Predators and Competitors

The oyster predators and competitors are crabs' sea stars, molluscs and organisms which grow on the shell and smother them. Korringa (1976) lists several species of crabs, fishes and molluscs as the predators of oysters. Some of these organisms such as crabs and fishes prey upon oysters weighing less than 5 gm.

Predatory gastropods such as *Busycon*, *Urosalpinx*, *Thais* sp. and *Trilonalia* attack young oysters along Atlantic and Pacific coasts. *Repana* sp. forms one of the serious oyster drills in Japanese waters. Among gastropods, *Cymatium cingulatum* has been observed to cause considerable damage to young *Crassostrea madrasensis* of size 35-55 mm in Tuticorin Bay in Gulf of Mannar (Thangavelu and Muthiah, 1983).

Among other major predators, the sea stars cause most difficult problem to the oyster growers in summer months. Fishes such as *Myliobatis* and *Pasgrus* have been observed as predators of oysters in France.

Polydora, a polychaete has been often found to cause noticeable damage to the oysters. The worm first settles on the shell and slowly occupies a position between the mantle and shell edges. It accumulates a mass of mud around itself and the oyster responds by secreting shell to cover the mud-worm complex. The meat of oysters heavily infested by *Polydora* is in poor condition and the shellfish are more susceptible to disease (Korringa, 1952).

Certain organisms such as Bryozoans (*Membranipora*), barnacles (*Balanus*) and mussels compete for space during settlement. Sometimes, the spat collectors are fouled by these organisms. On the fouled spat collectors, the larvae of oysters may not settle. To a large extent, the barnacles feed on oyster larvae. To avoid this, the spat collectors are laid precisely at the time when settlement of oyster larvae is likely to occur. Placing of spat collectors in advance before the spatfall would result in the fouling of collectors and thereby the setting of oyster spat is prevented.

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6

BIOLOGY OF *CRASSOSTREA MADRASENSIS* OF KAKINADA

K. A. NARASIMHAM[1]

INTRODUCTION

The concrete banks of the Kakinada canal harbour the oyster *Crassostrea madrasensis* (Preston) and they are particularly abundant at the mouth of the canal which extends and opens into the Kakinada Bay. The oyster beds are spread over an area of 2.25 ha and the oysters are regularly collected by the fishermen. Based on a survey conducted in April 1979, Narasimham *et. al.* (1984) estimated the density of the oysters in these beds at 112-1862 no/m^2 and the total stock at 90 t. There was no information available on the biology of *C. madrasensis* from the Kakinada Bay.

MATERIAL AND METHODS

During April 1985—July 1986, fortnightly samples of 60-250 oysters occurring in the subtidal region were collected at random from the oyster bed situated on the left bank (canal side) of the Kakinada canal. Also fortnightly water samples were collected from the oyster bed to study the temperature, salinity and dissolved oxygen. In the laboratory oysters were cleaned of all epifauna and epiflora with a wire brush and washed. Height was measured with a vernier calipers in the dorso-ventral axis, length in the antero-posterior direction and width from side to side in the broadest region when the valves were closed. The flesh was preserved in 5% formalin and examined about two weeks later. In the preserved oysters the sex and maturity stages were determined by examining the gonad smear under the microscope. The categorisation of the maturity stages was made following Ropes (1968). The gonad smears were studied in 20-25 oysters from each collection and a total of 760 oysters, of height range 37.5-95.2 mm were examined. Also histological preparations of gonad were studied in 120 specimens of height range 40.2-92.3 mm. Sections of 7-10 μ thickness were cut and stained in Delafield's haematoxylin and eosin. A test of variance for homogeneity was applied to examine whether the monthly sex ratio is uniform. The monthly sex ratio data were also analysed with the help of Chi-square test to see whether the sex ratio is different from 1 : 1 ratio.

The condition index was studied in 20 oysters from each collection and the overall height range was 25.0-94.4 mm. The flesh of each specimen was exposed to atmosphere for 1 h and then the still remaining moisture was removed with a blotting paper and then weighed. The dry flesh weight was recorded after keeping the wet flesh in hot air oven for 24 h at 90°C ± 1°C. The condition index was calculated as percentage of dry flesh weight in wet flesh weight.

Age and growth was studied by height-frequency analysis of 4,597 oysters measuring 6.0-102.7 mm. The oysters were grouped in 4 mm class intervals. To study the relationship between height and other body measurements the regression equation of the type $Y = a + bX$ was fitted by the least squares technique (Snedecor and Cochran, 1967). Where required logarithmic transformation was applied. A total of 64 specimens of height range 19.7-94.4 mm collected during April-June 1986 were used in these studies.

All linear measurements were taken to 0.1 mm accuracy, weight data above 5 g to the nearest 0.01 g and weight data of 5 g and below to the nearest mg. For various parameters the fortnightly samples were pooled and analysed on monthly basis.

[1] *Present address :* CMFRI, Research Centre, Kakinada-533 002.

Results

(i) *Environmental conditions*

The monthly average temperature varied from 22.8°C in January, 1986 to 33.6°C in April, 1985 (Fig. 1). It was generally high in summer (April-May), low in winter (December-February) and moderate during the southwest and northeast monsoons (June-November). The salinity ranged from 3.49‰ in October, 1985 to 35.01‰ in May, 1986. It was high in summer, declined with the onset of southwest monsoon in June, reached low values in August-December as the southwest monsoon (June-October) was followed by the north-east monsoon (November-December); it began to rise in January to reach the peak in summer. The dissolved oxygen fluctuated between 1.80 ml/l in August, 1985 to 4.34 ml/l in April, 1985.

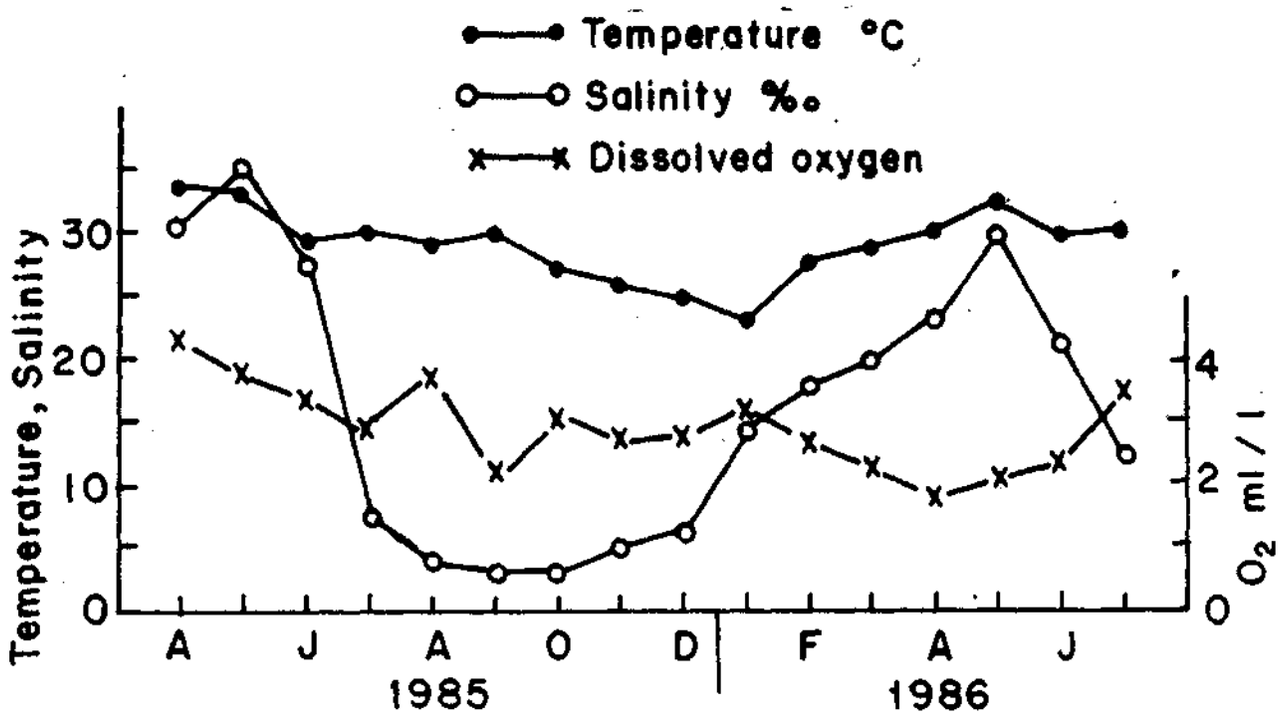

FIG. 1. Temperature, salinity and dissolved oxygen of the waters over the oyster bed in the Kakinada Bay.

(ii) *Maturity stages*

Active : The gonad is moderately developed and white in colour. Initially the follicles are few, small in size, mostly contracted and scattered in the connective tissue (Pl. I A). With further development they increase in number and are expanded. In males the follicles are packed with spermatocytes, spermatids and a few follicles may contain spermatozoa also (Pl. I B). In females oogonia appear at the periphery of the follicles, even before the gonads are completely empty. With further development the oocytes grow in size and are attached to the follicular wall by a stalk (Pl. II G).

Ripe : Gonad is well developed and cream coloured. Typically ripe gonads have dense appearence and the gametes are easily separated when punctured. In males the lumina of the follicles are densely packed with spermatozoa (Pl. I C). In females the follicles are packed with ripe ova which appear mostly free in the lumina. The ripe ova are polygonal or suboval in shape, measure 32-47 μ with nuclei 19-23 μ (Pl. II H).

Partially spawned : The gonad is pale brown in colour and the digestive gland is partly visible. When spawning is just initiated the gonad is well developed and with the progress in spawning it is reduced in size and becomes flabby. In both the sexes (Pl. I D, Pl. II I) there is a reduction in the number of gametes as they are discharged. This stage can also be called as spawning stage.

Spent/resting : In spent oysters the gonad is brown in colour and greatly shrunken. In both the sexes the residual gametes are present and connective tissue may be developing (Pl. I E, Pl. II J). In the resting phase the 'gonad' is well developed as it is filled with connective tissue and the oyster passes into indeterminate condition.

It was often observed that many follicles are not in the same level of development in the same specimen. For example in April-June, 1985 while majority of the follicles are in partially spawned stage others are mostly in active stage. In such cases the dominant maturity stage was assigned to the gonad.

(iii) *Spawning*

During April-June 1985, 80-95% of the oysters were in partially spawned stage indicating major spawning in these months (Fig. 2). In June most of the gametes were released. In July-October majority of the oysters (85-96%) were in spent/resting stage. Very few oysters (4-10%) were in partially spawned stage and among them most of the gametes were released. Gametogenesis was initiated in November and by December, 66% were in active, 26% in ripe and 8% in spawning condition.

In January-March, 1986 between 40 to 48% of oysters were in partially spawned stage; their number was reduced to 16% in April-May (Fig. 2). All the 4 maturity stages occurred in the above 5 months and their distribution suggests that the oysters undergo maturation and spawning more than once during these months; these months constitute the major spawning season. In June 2% and in July none of the oysters were in spawning stage and there was a progressive increase of spent/resting oysters in these two months indicating the completion of the reproductive cycle.

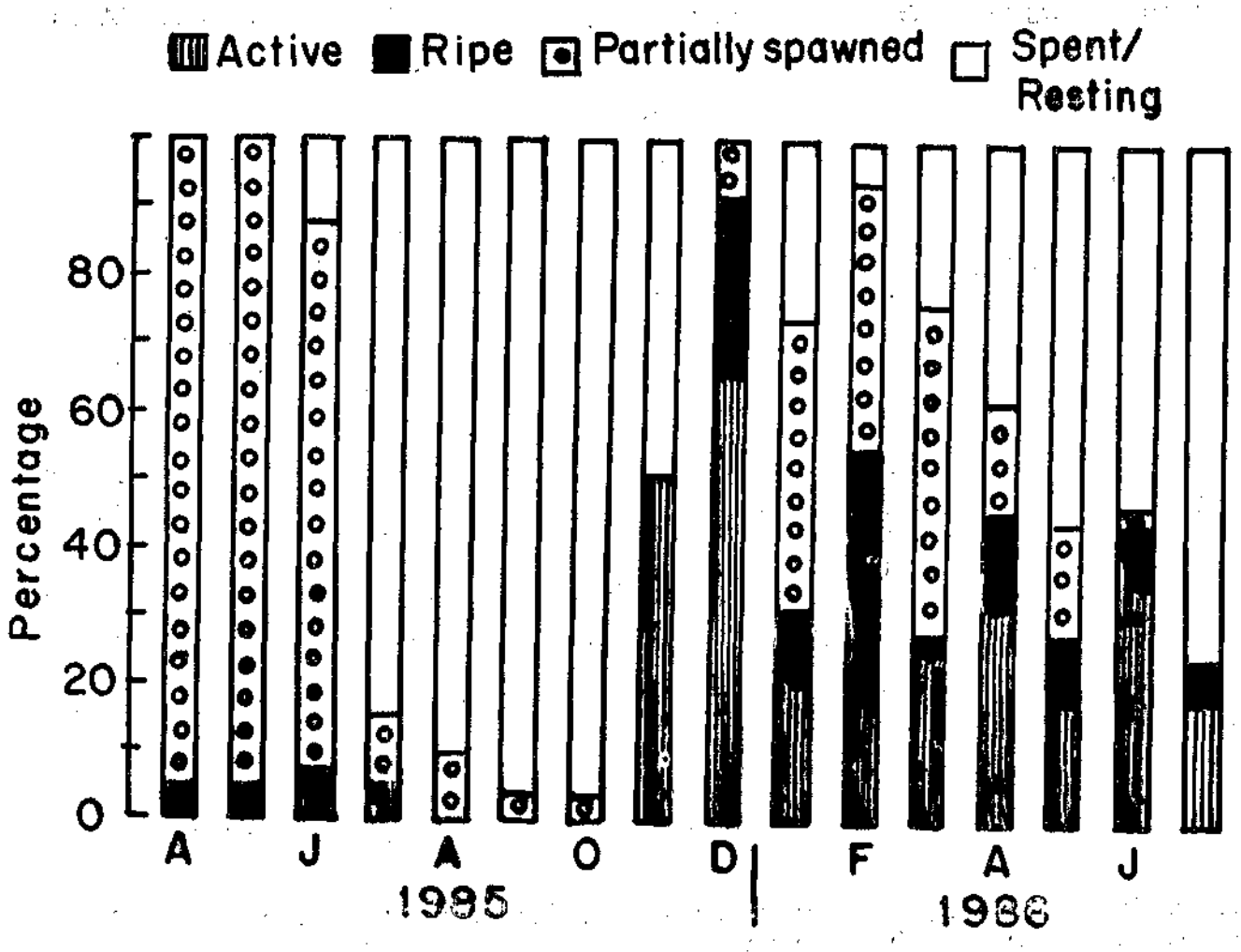

FIG. 2. Percentage occurrence of different maturity stages in *C. madrasensis*

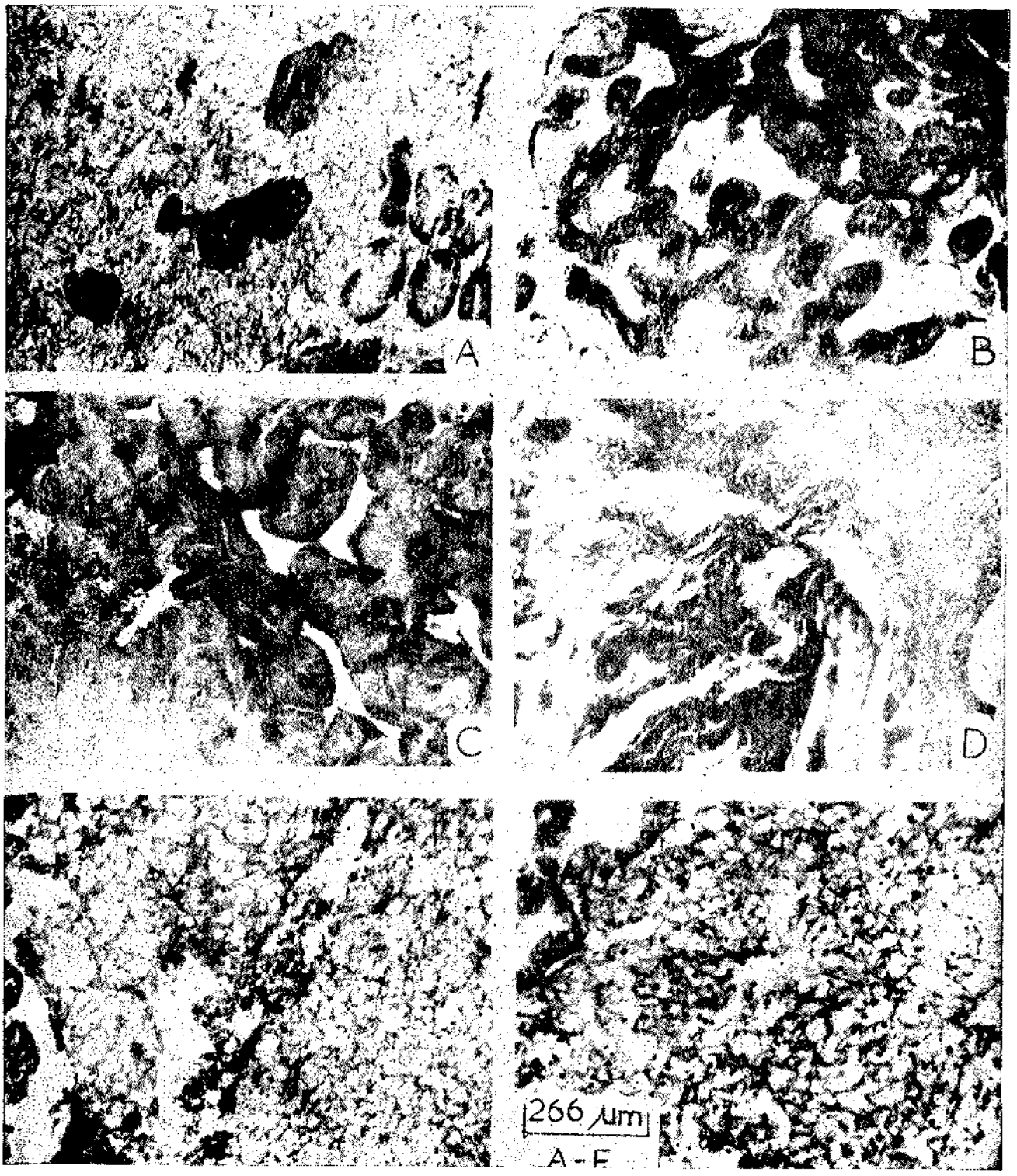

Plate I. A. Male gonad in early active stage showing a few follicles surrounded by connected tissue. B. Male gonad in active stage with many follicles and less of connective tissue. C. Male gonad in ripe stage. The follicles are expanded and densely packed. D. Male gonad in partially spawned stage. E. Male gonad in spent stage. Residual spermatozoa are present in the centre and the connective tissue is well developed. F. Resting condition. The connective tissue is well developed as the oyster passed into indeterminate phase.

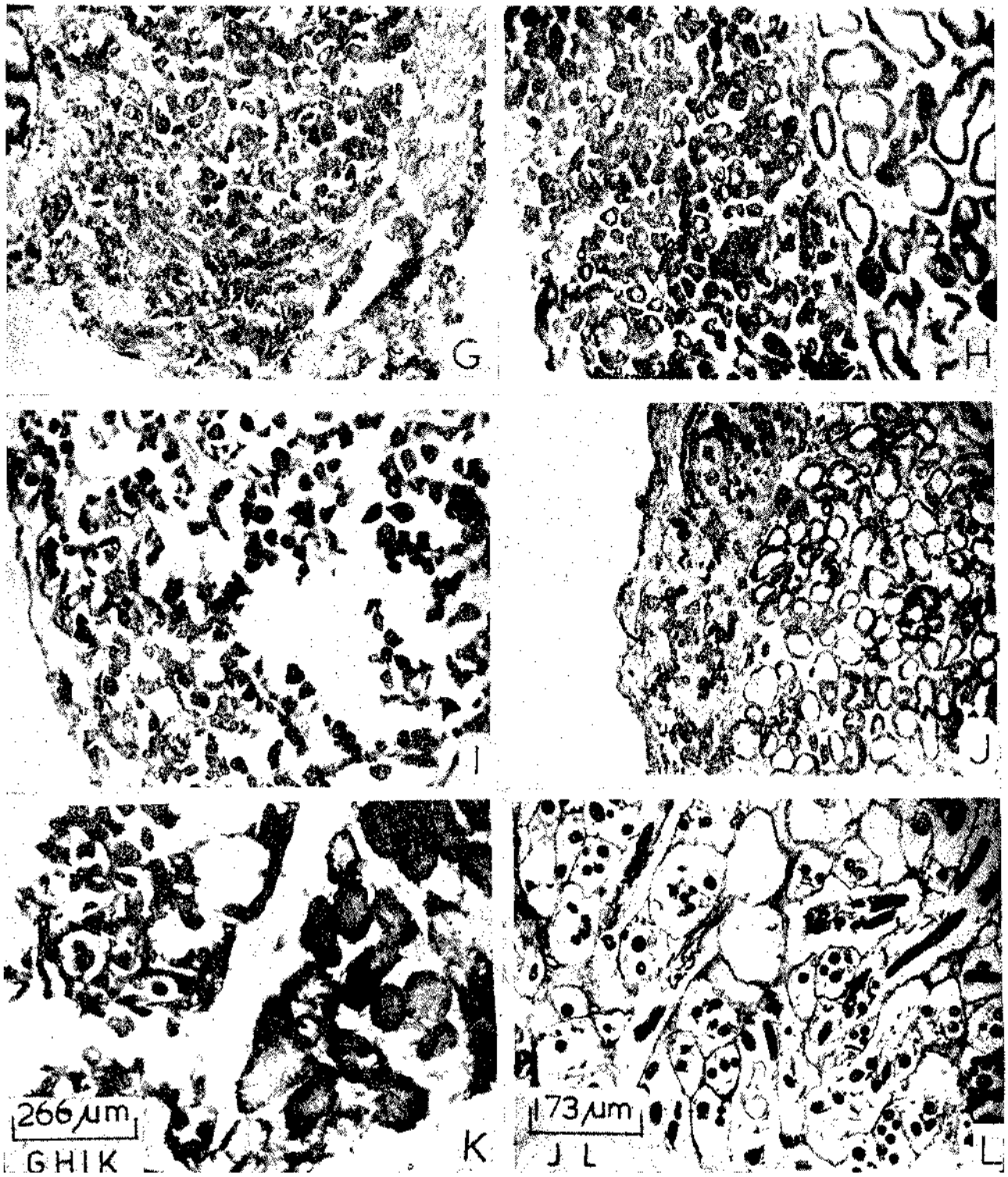

Plate II. G. Female gonad in active stage. The ova development is not uniform and the ova at top are far advanced in development than those below. H. Female gonad in ripe condition. The connective tissue is much reduced and many ova appear free. On the right is the digestive gland. I. Female in partially spawned stage showing some empty follicles. J. Shrunken gonad of a spent female. On the left are residual eggs which will be absorbed and on the right the digestive gland. K. Hermaphrodite showing ova on the left side and developing male follicles on the right side. L. Ovary infected by the bucephalid. Few eggs are present in the follicles.

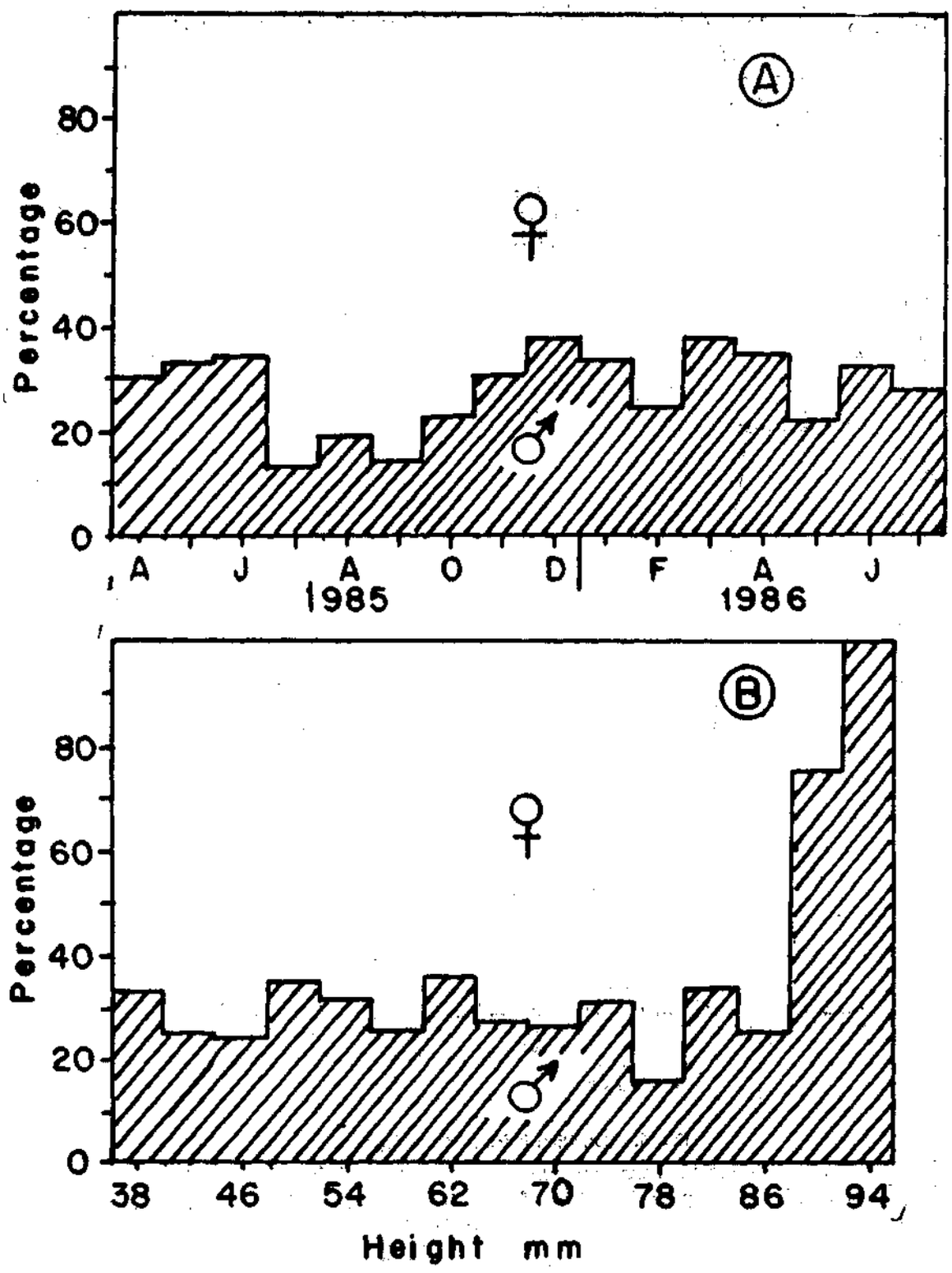

FIG. 3. Sex ratio in different months (A) and in different length groups (B) in *C. madrasensis*.

(*iv*) *Sex ratio*

Among the oysters which could be sexed, females invariably outnumbered the males in all the months and they formed between 62-87.5% during different months (Fig. 3A). The test of variance for homogeneity gave a Chi-square value of 17.78 (d.f = 1, 15) which is not significant at 5% probability. It was next ascertained whether the observed monthly sex ratio showed significant difference from the theoritical 1 : 1 ratio. The Chi-square values showed that at 5% probability there was a significant departure from the expected 1 : 1 ratio in all the months except in December, 1985 and March, 1986. It is concluded that the proportion of females is significantly higher than that of males.

The sex ratio in relation to height (Fig. 3 B) revealed that females outnumbered males in all the height groups excepting in 90 and 94 mm groups. However, the number of oysters examined in these two length groups were very few namely 4 and 1 respectively. The indeterminate oysters occurred in a few months and they formed 40%, 16%, 30%, 20%, 2%, 6.5% and 42% during July-November, 85, June, 86 and July, 86 respectively. During the study period males formed 24.3%, females 60.7%, indeterminates 9.5% and hermaphrodites 5.5%. Among the oysters which could be sexed the overall male : female ratio was 1 : 2.5.

(*v*) *Hermaphroditism*

Hermaphroditism was rare and 5% of the oysters were observed to the hermaphrodites (Pl. II K).

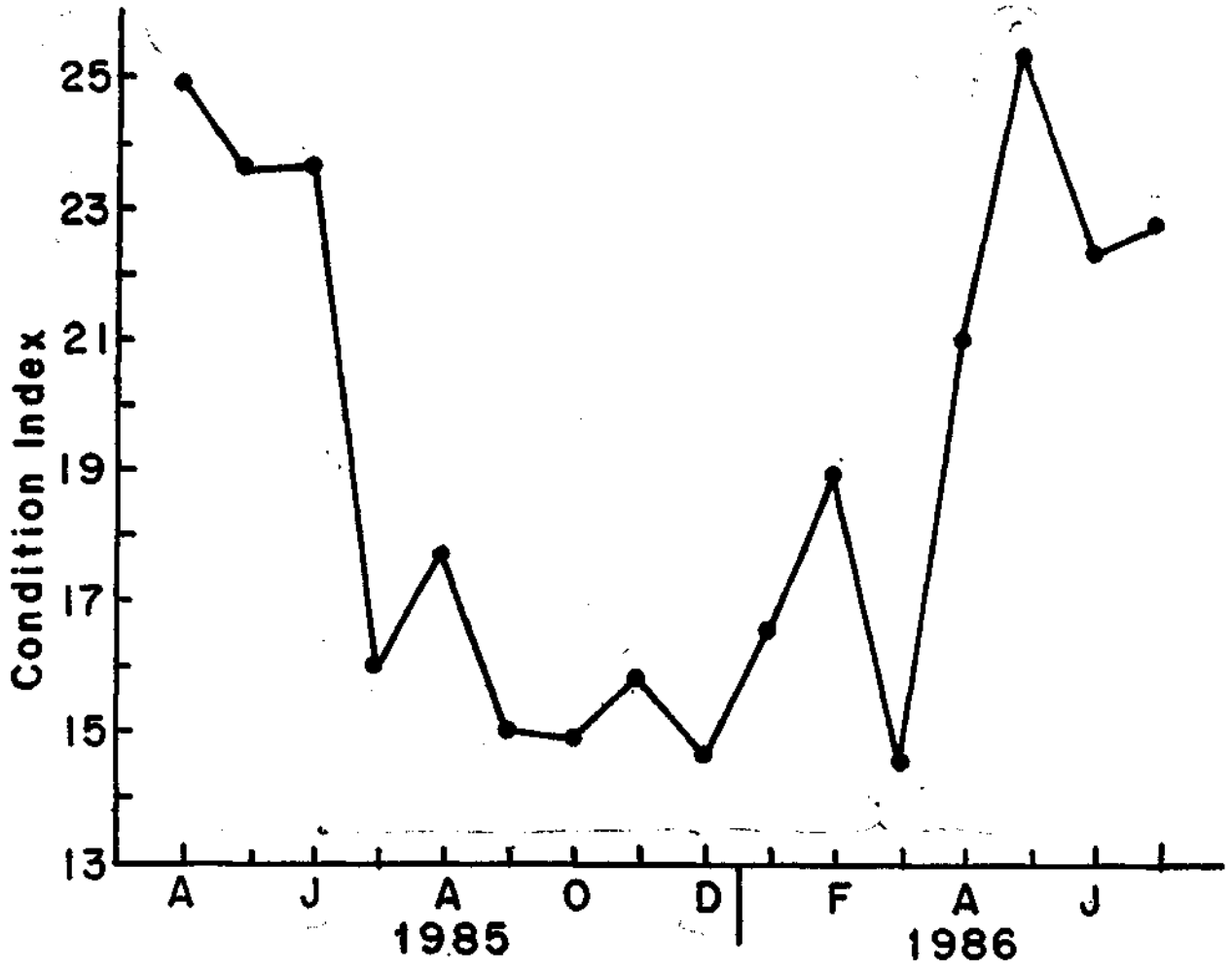

FIG. 4. Condition index in *C. madrasensis*.

(*vi*) *Pea crab infestation*

Two instances of infestation of the oysters by the pea crab *Pinnotheres* spp. were observed out of 840 oysters examined.

(*vii*) *Parasites*

Out of a total of 840 oysters examined the gonad of a single specimen was found to be infested by an unidenti-

fied bucephalid (Pl. II L). The gonad was distended with few eggs and the parasite invaded the digestive gland also.

(viii) Condition index

The monthly average condition varied from 14.5 in March, 1986 to 25.4 in May, 1986 (Fig. 4). It was high and ranged from 21.0 to 25.4 during April-June, 1985 and April-July 1986. There were two minor peaks in August 85 and February 86. In the remaining months it was low.

(ix) Age and growth

Mode A at 22 mm in April, 1985 (Fig. 5) was traced to 54 mm in August and there was no further progress till January when it shifted to 62 mm. By February, 1986 it moved to 66 mm and by May, 1986 to 70 mm. The oysters spawn during January-June and this mode can be taken to be the result of spawning early during the season i.e. January, 1985. Then it follows that in 6, 12 and 16 months the oysters attained 50, 62 and 70 mm height respectively. Mode B at 50 mm in April, 1985 progressed to 66 mm in July, 70 mm in November-December and to 74 mm in February, 1986. By taking the mode as the result of spawning late in the season i.e., in May-June, 1984, the oysters attained 58-66 mm height in one year and 70 mm in 20 months. Here also the modal progression was very slow during July-December. It is reasonable to consider that mode C at 22 mm in March, 1986 to be the result of spawning in January of the same year and in July, when 6 months old, the oysters attained 50 mm modal height. This study reveals that (1) the oysters grow fast during January-June and there was near cessation of growth during the second half of the year (2) a single year class may be represented by 2 broods and (3) in one year the oysters attain from 58 to 66 mm height.

(x) Height-weight and morphometric relationships

The regression equations fitted for various parameters are given in Table 1. The correlation coefficient (r) is very high for the various height-weight relationships indicating the goodness of the fit. The t test showed (Table 1) that the regression coefficients which ranged from 2.4110 to 2.6678 in equations 1-3 are significantly different from 3 at 1% probability indicating that the increase in total weight, shell weight and wet meat weight in relation to height of the oyster does not follow the cube law and that the growth is allometric. Similarly the t test showed (Table 1) that the b values in the height-length and height-width relationships are significantly different from 1 at 1% probability indicating the allometric growth of these body proportions.

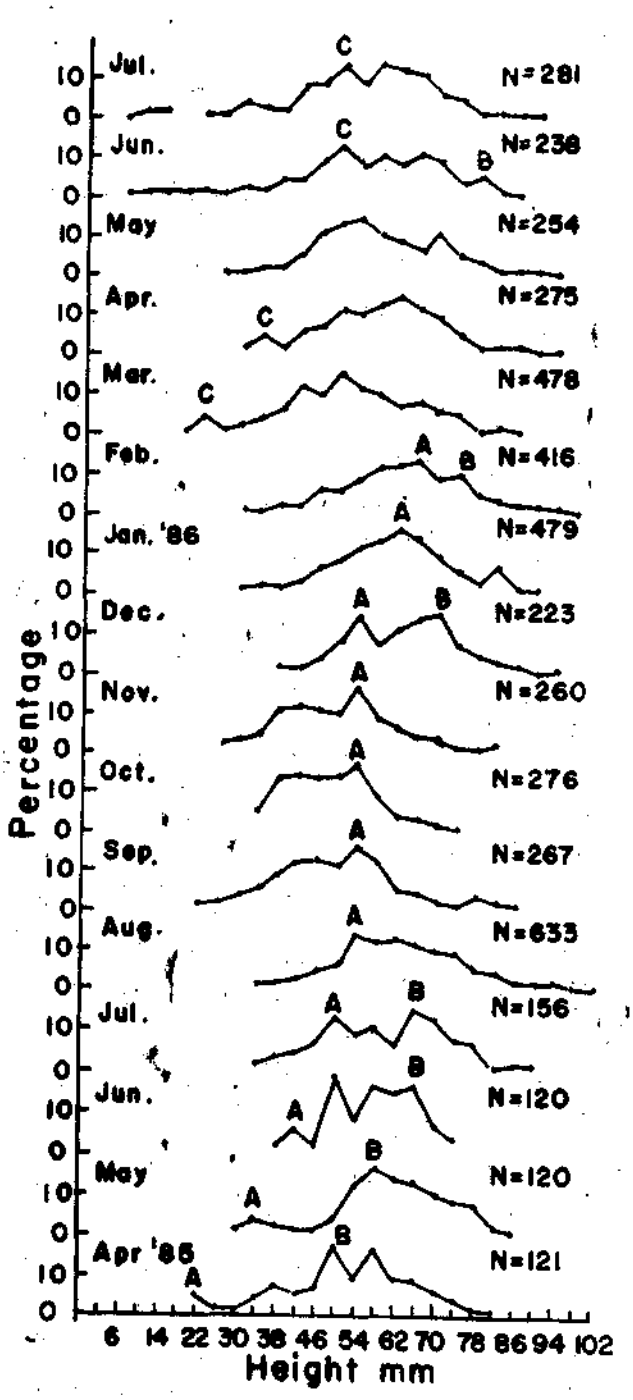

FIG. 5. Height frequency distribution in *C. madrasensis*.

TABLE 1. *Height-weight and morphometric relationships in* Crassostrea madrasensis. *Also the results of* t *test are given. The degrees of freedom is 62 for all the parameters.*

S. No.	Parameters		Equations		r	t
1.	Height (X) and total wt (Y)	..	Log Y=	−3.2421+2.6498 Log X	0.96	3.56
2.	Height (X) and shell wt (Y)	..	Log Y=	−3.3963+2.6678 Log X	0.95	3.13
3.	Height (X) and wet meat wt (Y)	..	Log Y=	−3.9245+2.4110 Log X	0.92	4.66
4.	Height (X) and length (Y)	..	Y=	7.5634+0.5823 X	0.84	8.74
5.	Height (X) and width (Y)	..	Y=	5.0008+0.2080 X	0.65	25.76

DISCUSSION

Earlier studies showed that in *C. madrasensis*, occurring under marine conditions, spawning takes place throughout the year (Paul 1942, Rao 1951) whereas in estuaries, either a restricted or biannual spawning was reported (Rao 1951, Rao 1974, Rao and Nayar 1956, Joseph and Madhystha 1982). In the present study, conducted in an estuarine environment, major spawning takes place during January-June. It is of interest to note that in the green mussel, *Perna viridis* also, which occurs on the oyster beds at Kakinada, major spawning takes place during January-May (Narasimham, 1980). Sastry (1979) reviewed the various exogenous and endogenous factors which influence the reproductive cycle in bivalves. Among them temperature and salinity received greater attention. At Kakinada, in both the oyster and green mussel, the major spawning period coincided with rising water temperatures and high salinities. During the non-spawning period, the temperatures were moderate and salinity was low. Joseph and Madhystha (1982) also observed that increase in salinity induces gametogenesis.

Rao (1956) observed that in *C. madrasensis* from Ennore backwaters males were dominant in January-June and females in the remaining months. However, in the present study the proportion of females was significantly higher than that of males in all the months and also in different length length groups.

Hermaphroditism in this species was observed by Rao (1953, 1956), Rao (1974) and Joseph and Madhystha (1982) and it was considered as a transitional stage to sex reversal (Rao, 1953). In this study also hermaphrodite oysters were encountered and they formed 5%.

There seems to be no other record except the present study regarding the infestation of *C. madrasensis* by the pea crab *Pinnotheres* spp. However, *C. gryphoides* and *C. cucullata* from Bombay showed pea crab infestation (Durve 1965, Awati and Rai 1931).

Infestation of *C. madrasensis* by bucephalid parasite seems to be rare in Indian waters, since apart from the present study, Samuel (1978) reported from Tuticorin.

In this study the condition index (CI) was high during April-June, 1985 when majority of the oysters were in partially spawned stage and with the completion of spawning it fell in July; it remained low during August-December when spent/resting oysters were dominant. In March majority of the gametes were released and the CI declined and with the progress of another reproductive cycle during April the CI was high. In June-July the majority of the oysters were in spent/resting stage and the considerably high CI in these months may not be related to the reproductive cycle. Rao (1956) also observed the fall in CI in this species following spawning.

Rao and Nayar (1956) observed that in the Adyar estuary *C. madrasensis* attains 50.6 mm height in 13 months. In the Bhimunipatnam backwaters Reuben *et al.* (1980) found that this species grows to 80 mm in one year; in the Mulky estuary it attains 91.5 mm in first year and 142 mm in second year (Joseph and Joseph, 1985) and in the Vellar estuary 48.81 mm, 84.97 mm and 111.7mm at ages 1-3 respectively (Somasekar *et al.*, 1982). In the Kakinada Bay this species attains 58-66 mm in one year. Thus considerable disparity in growth is discernible at different places. In the present study growth was slow during June-December when the salinities were very low. Similar observation was made by Rao and Nayar (1956) on this species.

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7

TECHNIQUES OF COLLECTION OF OYSTER SPAT FOR FARMING

P. MUTHIAH [1]

Methods of spat collection commonly practised:

Dependable and consistent supply of seed is an essential prerequisite for viable aquaculture industry. Oyster seed commonly called *spat* is procured from natural resources by placing suitable cultch materials in the water column at the most appropriate time and place. Cultches used vary from twigs of various trees to modern synthetic materials. In 1670, Goroemon Kobayashi of Hiroshima first used bamboo poles with twigs and nets for successful collection of oyster seed. Bamboo branches and branches of trees such as oak, pine and chestnut were also used. Shells of scallop and oysters are commonly being used in the form of ' rens ', hung horizontally or vertically from the racks. Recently *Netlon* a hexagonal plastic netting with a mesh size of 5 cm. of size 5-10 × 15 cm. is also being used for spat collection (Fujiya 1970, Bardach *et al.* 1972). In New Zealand Curtain (1974) found that asbestos cement strips or sticks serve most effectively in spat collection. In Carribean bitumen painted wooden planks and branches of red mangrove are used for collecting seed oysters. Plastic mesh collectors similar to *Netlon* are used in Canada and U.S.A. (California, Virginia). Ropes intertwined with twigs are employed in Italy. Rens of shells, metal net baskets or triangular wooden frames with empty shells inside serve as spat collectors in North America (Imai, 1971). Thomson (1954) states that sticks of black and orange mangrove, wattle, white cypress and swamp oak and brush woods in addition to Fibro cement slates or tarred sawn timber slates are used as spat collectors in Australia. Quayle and Smith (1976) state that in British Columbia though rens of oyster shells are commonly used, strips of wood veneer of spruce or fir that are coated with cement are also popular. In France the main spat collector is ribbed PVC tubes 1.2 m long and 2.3 cm in diameter. Six tubes are tethered together as a bundle and placed on a 'table'. Rens (Chapelet) of oyster, mussel and scallop shells are also used. In flat oyster culture stacks of 10-12 lime coated tiles each arranged as a bouquet using a zinc wire are placed on a pole erected in the bay area for spat collection.

In India, Hornell (1916) experimented with lime coated tiles to procure oyster spat in Pulicat Lake. The choice of cultch material depends on culture methods adopted, material availability and economic and practical utility. In oyster culture experiments carried out at Tuticorin, a number of cultch materials were tried. This paper outlines the results of experiments on spat collection carried out at Tuticorin during 1978-80.

Experiments on spat collection:

Because of easy availability cultch materials i.e., oyster shells, coconut shells, asbestos sheets, mussel shells and tiles were tried during 1980 spawning season.

Oyster shell rens:

Holes were drilled at the centre of 2,000 oyster shells. 20 such shells were strung on a G.I. wire (No. 10) of length 1½ metres. Each unit is called a ' ren '. Since the depth of water in the bay area was 0.5 to 1.5 m, 100 rens were laid horizontally on a rack (Pl. I a). The number of oyster spat collected on oyster shells ranged from 0 to 27 and average spat/shell was 7.

Coconut shells:

1,200 empty coconut shells dipped in tar and dried were punctured at the centre. A unit of seven such shells were strung on a nylon rope (3 mm) of length

[1] *Present address*: CMFRI, Research Centre, Tuticorin-628 001.

1½ metres with interspace of 10 cm. Thus 180 rens having a surface area 75 m² were placed horizontally on a rack (Pl. I b). Maximum number of spat settled on a coconut shell was 5. Average settlement was 1/shell.

Asbestos sheet :

Lime coated asbestos sheets (100 nos.) each of size 120 × 80 cm were placed in grooves of pamlyra reapers which were tied to poles driven in the bottom (Pl. I c). Minimum and Maximum settlement were 2 and 10 respectively. Average settlement per sheet was 4.

Mussel shells :

Several strings of mussel shells were placed horizontally on a rack. The spat settlement was moderate. The average spat settlement was 5/shell.

Velon screen and polythene liner sheet :

Other spat collectors used in experiments are velon screens, polythene liner sheets and PVC tubes. Velon screen or polythene liner sheets of size 4.25 × 16.5 cm encircled on a metal frame (Pl. I d) were tied to the rack. Spat settlement was 87 and 215 respectively on the velon screen and polythene liner sheet. Removal of spat from these collectors was easier than scraping of spat from tiles.

PVC tubes :

Nine PVC tubes of 3 cm dia. and length 1 metre fixed on to wooden rings at either end. About 20 such rolls of these PVC tubes were laid on a rack. Average settlement of spat was 30/single tube.

Tiles :

Lime coated tiles 24 × 15 cm in size were kept in nylon knitted cages each of 100 × 90 × 15 cm size, in such a way that the concave side of the tile faced downwards. Fifty tiles in 2 tiers of 25 each were placed in each cage (Pl. II a). A total of twenty cages were arranged on a rack (Pl. II b). Settlement on the tiles was encouraging. In some tiles as many as 120 spat settled down. The average settlement of spat/tile was 33.5 (Pl. II d).

Spat Collection :

Based on the encouraging results obtained, large-scale collection of oyster seed was attempted using lime coated tiles as collectors. Thangavelu and Sundaram (1983) have given details of the lime coating process. Lime coated tiles numbering 18,000 to 50,000 were laid during the peak spawning seasons i.e., April-May and August-September from 1978 onwards upto 1984 and the details are given in Table 1.

TABLE 1. *Details of Spat Collection*

Items	Year	1978	1978	1979			1979	1980			1980
	Season	April-May	August-Sept.	April-May			August-Sept.	April-May			August-Sept.
	Place	Creek	Creek	Creek	Natural bed	Tuticorin Bay	Tuticorin Bay	Creek	Natural bed	Tuticorin Bay	
Total No. of racks		30	30	10	20	18	20	2	2	30	Not tried
Total Number of lime coated tiles provided		27,000	30,000	10,000	20,000	18,000	20,000	2,000	2,000	30,000	
Number of spat settled/m² of surface area ..		153	Nil	76.9	92.3	316	43.8	100	107.7	184.1	
Total spat collected ..		3,00,000	Nil	50,000	1,20,000	3,90,000	57,000	13,000	14,000	3,59,000	

In 1978 April-May season 30,000 lime coated tiles were provided in the creek and 30,000 spat were collected. Number of spat per m² of surface area of cultch was 153. During August-September season in the same area, there was no settlement because of the closure of creek mouth.

In 1979, April-May season spat collection was carried out in three different places viz., creek, natural bed and bay area (Pl. II c). The spat collection in the bay area was very good. The number of spat/m² of the surface area of cultch was high (316/m²) in the bay

when compared with creek (76.9/m²) and natural bed (92.3/m²), thus indicating, the shallow intertidal open sea area is better suited for attempting large-scale spat procurement and its great potentialities. In August, 1979, however, in the same area 57,000 spat could be collected.

Experimental collections made in 1980 April-May season also followed an identical pattern of 1979 results. Therefore from 1981, spat collection was intensified in April-May season. The average spat settled per tile during 1981, 1982, 1983 and 1984 were 12, 15.6, 29 and 15 respectively.

Oyster shell rens were also employed in the April-May spawning seasons from 1980 onwards. The average spat/shell ranged from 5.8 to 6.5.

Setting behaviour :

In order to place the cultch at the appropriate time and position, precise information is essential on the setting behaviour of oyster larvae. Very often strong currents interfere with the larval settlement. Heavy collection of 386/m² in the bay area may be due to its shelterd nature. Korringa (1952) points out the concentration of setting in the periods of slack water.

Hornell (1916) recorded better spat settlement on the lower or concave surface than the convex surface of the tile. A maximum of 80 and minimum of 9 spat were found attached to the concave side. The ratio of spat settled on concave and convex surfaces was 5: 1 (Silas *et al.*, 1982). Average settlements on the concave and convex sides were 21.1 and 2.6 respectively. As indicated by Knight Jones (1951) for Essex oyster beds, there appears to be a preference to the darker lower surface than the convex surface which is laid with silt. Other factors that are known to influence the setting behaviour of oyster are the intensity of light, depth, angle of surface, cleanliness and roughness of the surface (Quayle, 1980).

Spatfall prediction :

For successful spat collection, timing for laying of cultch materials is one of the crucial factors. Therefore, it is very essential to determine accurately the time of spatfall. If spat collectors are placed well in advance of spatfall they may be coated with a thick film of silt and fouled becoming unsuitable for the larvae to settle down. Studying the gonadial condition of oyster and concentration of the oyster larvae in plankton will help to determine the time for placing the spat collectors. Matthiessen (1974) stated that the suspension of strings was postponed until the density of larvae reached 25 larvae/litre. During the peak spawning season i.e. April-May and also in August-September when 70% of female oysters observed were in fully ripe condition, spat collectors, oyster shell rens or lime coated tiles would be kept ready for laying. With the appearance of the oyster larvae in the plankton samples collected from the area, actual laying of spat collectors would start. The peak occurrence of oyster larvae was noticed to be in the first fortnight of April when average salinity was 33.8‰. and the temperature ranged from 30 to 31.5°C. Rao (1951) also found identical conditions in the oyster beds at Madras.

Foulers on the cultches :

Mackenzie (1970) states that the intensity of fouling can be reduced by delaying planting of spat collectors until larvae were ready to settle. Loosanoff (1961) was of the opinion that fouling intensity was less in the polystream (polychlorinated benzene) treated cultches and the number of spat settled was three times more than that on the untreated ones. The spat settled on tiles were kept in the cages for two months before scraping of spat was done for further rearing. During the period the common foulers noticed were barnacles, serpulids, *Anomia*, *Polycarpa* sp., *Styela* sp. and compound ascidians. These were periodically cleaned.

Hardening :

Seed oysters to be transported have to be conditioned. Hardening is a process by which these seed are periodically exposed during low tides. Experiments conducted on hardened and non-hardened seed, showed that the hardened seed could be kept in semiarid condition upto 120 hrs. with 76% survival. The survival rate of non-hardened seed for the same period is 22%, thus indicating the ability of the hardened seed to withstand semiarid or arid conditions better.

Remarks

Condition of cultches and the method of exposure are the prime factors for spat collection. The cultch materials must be finely roughened, free from slime and without any secretion such as gums and resins. Most of all the spat collectors must retain the oysters until they are scraped for further rearing or until they attain marketable size. They should withstand the wave action and the attack of boring organisms.

Spreading cultch materials on the bottom, or placing them on racks or suspending from raft are the three methods of exposure of spat collectors in the water column. Raft suspension method is efficient but the cost of flotation is high. Efficiency of rack system for

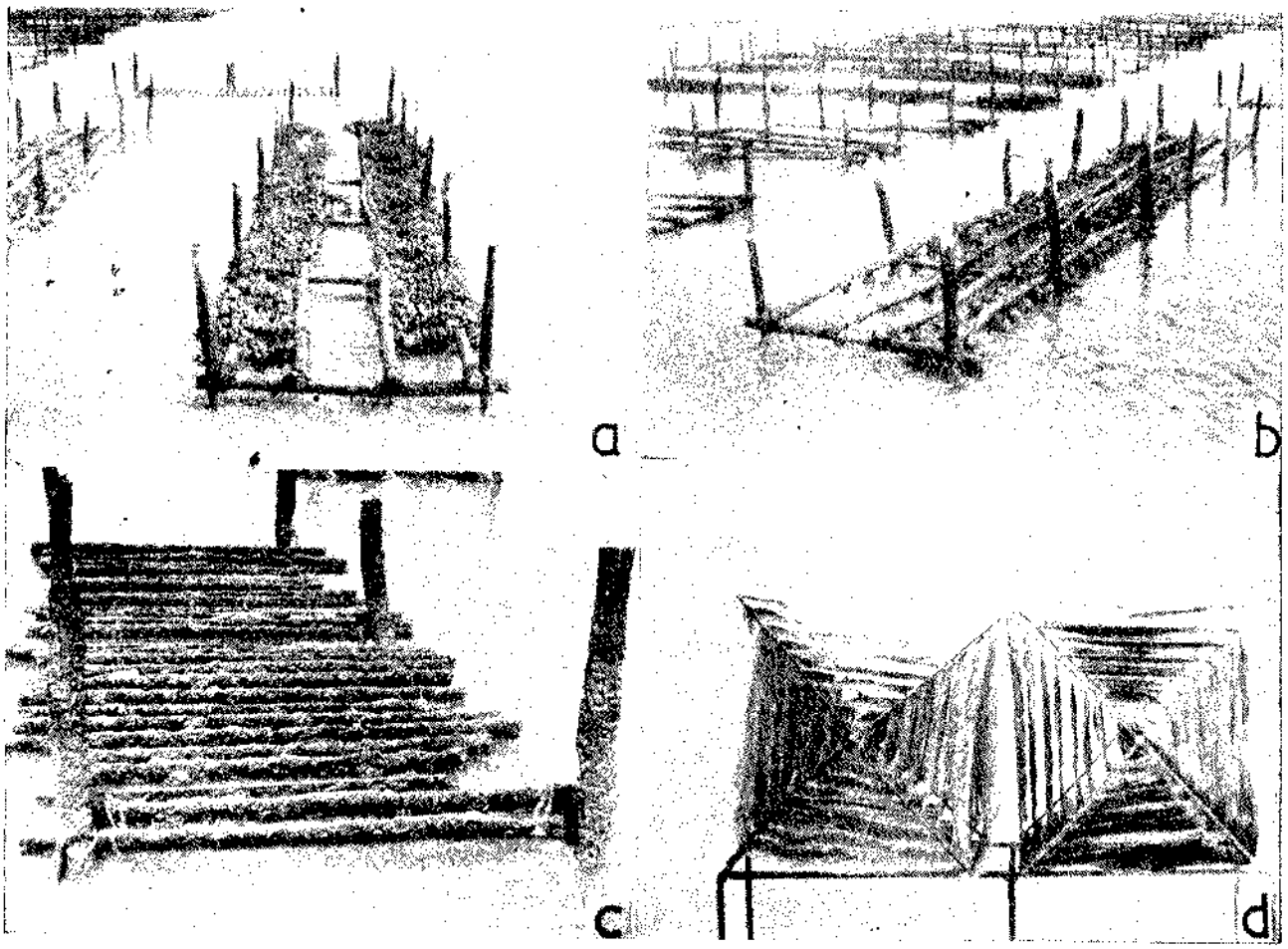

PLATE 1. (*a*) Oyster shell rens on a rack for spat collection. (*b*) Coconut shell rens on a rack. (*c*) Asbestos sheet. (*d*) Velon screen and polythene liner sheet encircled on a metal frame used for oyster spat collection.

spat collection is better because of lesser attack by predators and accomplishment of recovery of cultch materials.

Of the various materials of spat collectors, lime coated tile is being used in Holland and France for commercial spat collection. The advantage with this spat collector is that the tiles used in one season can be recycled for subsequent season. In the areas where sufficient depth is there to rear the oysters by suspension method, rens of oyster shells can be suitable material for spat collection. Recent uses of PVC pipes and synthetic materials for spat collection will help in easy removal of spat instead of labour involving scraping process.

Although a few lakhs of spat were collected during the experiments at Tuticorin, it is possible to procure more by employing larger number of suitable spat collectors. But natural setting may vary with location and time, and is largely governed by year to year variations in the environmental characteristics. Hence dependence on nature alone for seed requirements has its limitations. Hatchery production of seed appears to be the assured means of continual, dependable supply for culture operations. Considering the high cost that may be involved in the hatchery production of seed in advanced countries, it appears that in tropical countries like ours the running cost of the hatchcery operations may not be prohibitive. Glude (1979) reported that demand for edible oysters may increase to more than 2 million tons by the year 2000 A.D. By resorting to hatchery production of seed oysters, it is possible to set up 'seed banks' as tried in the Pacific Northwest coast (Glude, 1979) for effecting regular supply to oyster farmers.

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8

PRODUCTION OF OYSTER SEED IN A HATCHERY SYSTEM

K. NAGAPPAN NAYAR[1], K. SATYANARAYANA RAO[1], M. E. RAJAPANDIAN[1], C. P. GOPINATHAN[1] AND A. D. GANDHI[1]

INTRODUCTION

Although oyster culture dates back to First century B.C., the development of hatchery techniques for the production of oyster seed on a year round basis is a recent innovation. Since early 1950s attempts for large scale production of oyster seed have been initiated. Loosanoff and Davis (1952), Dupuy *et al.* (1977) and AQUACOP (1977) have successfully produced seed of the oysters *Ostrea edulis*, *Crassostrea virginica* and *Crassostrea gigas*. Nayar *et al.* (1984) have successfully accomplished the production of seed of *C. madrasensis* on a large scale at the molluscan hatchery laboratory of Central Marine Fisheries Research Institute at Tuticorin. At this hatchery cultched as well as free or cultchless spat are produced. The production of oyster seed by hatchery techniques is accomplished in six phases of operations viz., (1) Conditioning adult oysters for maturation of gonads (2) Induced spawning (3) Larval rearing (4) Culture of algal food (5) Preparation of spat collectors and (6) Setting of spat. These six functions although interrelated are independent phases of operation and easy to follow and implement.

CONDITIONING OF ADULT OYSTERS FOR MATURATION OF GONADS

The aim of conditioning oysters is to induce maturation of gonads rapidly by feeding the oysters with a rich supply of food which provides adequate nutrition required for gamatogenesis and build up of the reproductive organs.

Selection of broodstock

The oysters for broodstock are selected keeping in view the area, growth and condition factor, size and age of the standing oyster population. They are collected from population in areas where they are known to occur in healthy condition. The oysters chosen for conditioning are collected from areas where the salinity regime is comparable to that in hatchery. Otherwise the oysters have to be acclimatised before they are conditioned. The prevailing temperature of the collection area has to be recorded first, since on this basis manipulation of temperature regime is effected for conditioning of the oysters for maturation and induced spawning.

Oysters of the size (length) range 60 mm to 90 mm are ideal and 30% of this should be of 'O' year class or just one year old (60-75 mm) in order to be assured of the availability of males in the broodstock. Before stocking the oysters a smaple of 10 numbers are opened to ascertain whether they exhibit uniform maturity stage, preferably immature or spent. If there are more than one stage the conditioning cannot be done effectively. A minimum of 500 oysters are kept as broodstock in the hatchery.

The selected oysters, 25 in each batch are cleaned thoroughly and placed on a synthetic twine knit P.V.C. frame in a 100 litre fibreglass tank (75×50×25 cm) and raw seawater is filled in the tank and well aerated. The water level inside the tank is maintained at half the height of the tank and 15 litres of mixed phytoplankton cultured in outdoor tanks using fertilised medium (Gopinathan, 1982) are added twice during a day between 09.00 and 17.00 hours at 4 hours intervals. The phytoplankton diet is composed of diatoms such as *Chaetoceros affinis*, *Skeletonema costatum*, *Thalassiosira subtilis* and *Nitzchia closterium*, the phytoflagellates, *Isochrysis galbana* and *Pavlova* sp. and the microgreen alga *Chlorella salina*. On an average the cell concentration of the algae should be about 1.0 million cells/ml. The oysters are conditioned at

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001.

about 5°C below ambient temperature for 10 to 20 days. The tanks in which the oysters are reared are cleaned every day to remove dirt and faeces and filled with fresh raw seawater before commencing feeding.

On the tenth day a sample of three oysters are opened to observe the condition of the gonads. The right valve of the oyster is removed with the help of a shucking knife. By using a Pasteur pipette a few shallow cuts are made on the mantle surface, a small quantity of gonadal contents are drawn with the pipette, a drop of this is placed on a slide with a cover slip and examined under microscope to determine if the gonads of the oyster contain ripe sperms or ova. Rapid directional movement of the particles indicates sperms. The ripe ova of a mature ovary of *Crassostrea madrasensis* are pear shaped and compressed. The presence of spherical and subspherical ova in the sample indicates the ripeness or the readiness of the oyster to spawn. Such ova measure 50 to 60 μ in diameter. If the oysters are not mature they are conditioned for another five to ten days. The oysters conditioned to mature stage by this process are transferred to spawning tanks.

Induced Spawning

The conditioned oysters spawned on giving thermal stimulation by transferring them to water of higher temperature. Firstly the conditioned oysters are thoroughly washed and transferred to a 100 lit. Perspex spawning tank containing 50 l. of filtered seawater with temperature 2 to 4°C above the ambient temperature level. This is achieved by operating a heating element with porcelain coating which is controlled by a thermostat (Pl. I A). Proper aeration of seawater is provided in the tank. During the first hour the oysters stimulated thus commence spawning. If spawning cannot be achieved by this method, fresh sperms stripped from a sexually ripe male are introduced in the tank containing the broodstock.

The sperms from a stripped male (0.25 ml.) are diluted with 10 ml of filtered seawater. This suspension is drawn in a long pipette (10 ml.) which is immersed and placed above the oyster along the incurrent side and sperms are slowly released from the pipette. This stimulation should be given to all the oysters individually in the tank. At intervals of 10 to 15 minutes more sperm suspension are released till the oysters start spawning.

Once an oyster starts spawning, it is transferred to a three litre glass tray containing filtered seawater at ambient temperature. 15 to 20 numbers of such trays are kept ready for operation. Only one oyster is placed in each tray. Each individual oyster is allowed to complete its spawning in the tray. If the female is a heavy spawner and the water becomes highly milky it is transferred to another tray to complete spawning. The males spawn by continuous ejection of white stream of seminal fluid containing spermatozoa (Pl. I B) and the females by rhythmic ejection of ova at intervals. The spawning oysters in the glass trays are carefully watched and each removed as spawning is completed. This is done to prevent the oysters from filtering the gametes. The filtered seawater containing eggs (Pl. I C) from each spawning tray is poured through a 100 μ stainless seive or nylobolt seive into a 10 litre glass beaker. At this stage mild aeration is given to ensure sufficient supply of oxygen from air bubbles produced by an aquarium air stone.

Fertilization of Eggs and Estimation of Fertilized Eggs

The ova in each beaker are fertilized within 45 minutes after being spawned, from a pooled sperm suspension in the individual trays in which the males have spawned (Pl. I D).

50 ml. of a pooled sperm suspension obtained from as many males as possible is added to each beaker containing egg suspension. After 20 to 30 minutes a 2 ml sample of the fertilized eggs is taken and examined under microscope to ascertain the extent of fertilization. This can be determined by noting the formation of polar body. If 10% or more of these eggs do not exhibit this feature, then more sperm suspension (15-20 ml.) is added.

When fertilization is complete, the 10 l. beaker containing the fertilized eggs is filled with filtered seawater. After proper mixing of the eggs in the container a 1 ml. sample is drawn quickly with a blowout pipette. The sample is drawn quickly as the eggs have a tendency to settle out of suspension. If the concentration of the eggs is high the sample is made up to 10 ml and one ml of subsample is taken. The sample is then transferred to a counting chamber and the number of eggs counted with a compound microscope. After counting and recording the number of fertilized eggs/ml. will be multiplied by the dilution factor and the number of fertilized eggs in 10 l. is estimated.

At the end of one hour, aeration is suspended. The fertilized eggs settle at the bottom. After 15 minutes, the supernatant water, containing sperms, unfertilized eggs and debris is removed. Fresh seawater is added

and decanting is carried out 3 times. Finally 40 to 50 l. seawater is added to the tank and mild aeration is provided. At the end of 4 hours following cell divisions, the eggs attain the morula stage and begin to swim in the column layers (Pl. I E and F and Pl. II A and B). The larvae are allowed to develop for the next twenty hours.

Larval Rearing

The sequence of developmental stages and growth and rearing of larvae from the straight hinge to the eyed or pediveliger stage are described below.

Straight-hinge stage

The straight hinge or 'D' shell larval stage is attained at the end of 20 hours (Pl. II C) The larvae are semi-transparent with the velum protruding out and creating a strong ciliary current which directs minute particles of food into the stomodaeum. The larvae swim vigorously and some of the larvae show a slow circular movement under the microscope. The actively swimming larvae are separated by siphoning them from the tank, leaving the sluggish ones. This process of culling is continued during the first two days or till the entire stock of the larvae are of uniform size and movement. On an average, the larvae measured 66 μ in length on the first day. At this stage the larvae are reared in one ton fibreglass tank. The total number of the larvae is estimated and stocked at the density of 4 larvae per ml in the rearing tank. The larvae are fed with phytoflagellates at the end of 24 hr.

Umbo stage

On the third day, the larvae become slightly oval in shape and measure 100 μ. This stage is considered as early umbo stage. At this stage the larvae are filtered through 80 μ filter in order to segregate the smaller ones or the non-growing larvae.

On the 7th day, the umbo is seen distinctly and pronounced concentric rings are found on the shells as the larvae grow (Pl. II D). The larvae measure 150 μ at this stage.

In 12 to 15 days the 'late umbo' larvae measure between 260 and 270 μ. During the rearing period, on every third day the total number of larvae in each tank is estimated to determine the mortality rate. On an average, per day a larval mortality rate of 2% to 3% is normal. If the mortality rate is very high it means the conditions are unfavourable. In such a case the larvae should be filtered every day to remove the dead and non-growing larvae. Further it is advised to treat the larvae with specific doses of water soluble antibiotics such as Streptomycin sulphate or Chloramphenical.

The larvae are filtered out from the tank and immersed for 15 minutes in seawater containing the antibiotic Streptomycin sulphate at a strength of 50 PPM. This process of treatment controls to a large extent fungal diseases. The dipping of larvae in antibiotic medium may be given once or twice during the larval period if necessary.

Eyed stage

An irregular eye spot is observed between 13 and 17 days when the larvae grow to a size of 280 μ (Pl. II E). It becomes distinct at the size of 290 μ. The eye spot is present in the lower quadrant of the ventral region close to the right angle of the dorso-ventral and antero-posterior planes.

Pediveliger stage

Between the 14th and 18th day, the functional foot emerges. At this stage the larvae measure 330-350 μ (Pl. II F). At this stage they descend to the bottom and start crawling. This is known as 'swimming creeping' stage which has been designated as pediveliger by Carriker (1961). The spat start setting within 24 hours or sometimes setting is prolonged to 2 to 6 days depending on the availability of favourable substratum.

Spat

The pediveliger larvae settle down, losing the velum totally. The shell edges grow hexagonally and the larvae develop the characteristic adult features and metamorphose into the spat (Pl. III A). The young spat measures 450 μ. The eye spot is traceable at the stage and it disappears 24 hours after attachment.

Larval density

As the larvae grow their number per tank is reduced. It is absolutely necessary to reduce the number of larvae since the growing oyster larvae require greater space for optimal feeding and growth.

The larvae are transferred to 10l beakers using 100 μ mesh sieves. In the beaker they are mixed well and one ml is drawn and transferred to cell counter. The number of the larvae is counted under microscope and the total number is estimated. This study is carried out in the case of all individual containers. Measured quantities of the larval medium is added to the rearing tanks so

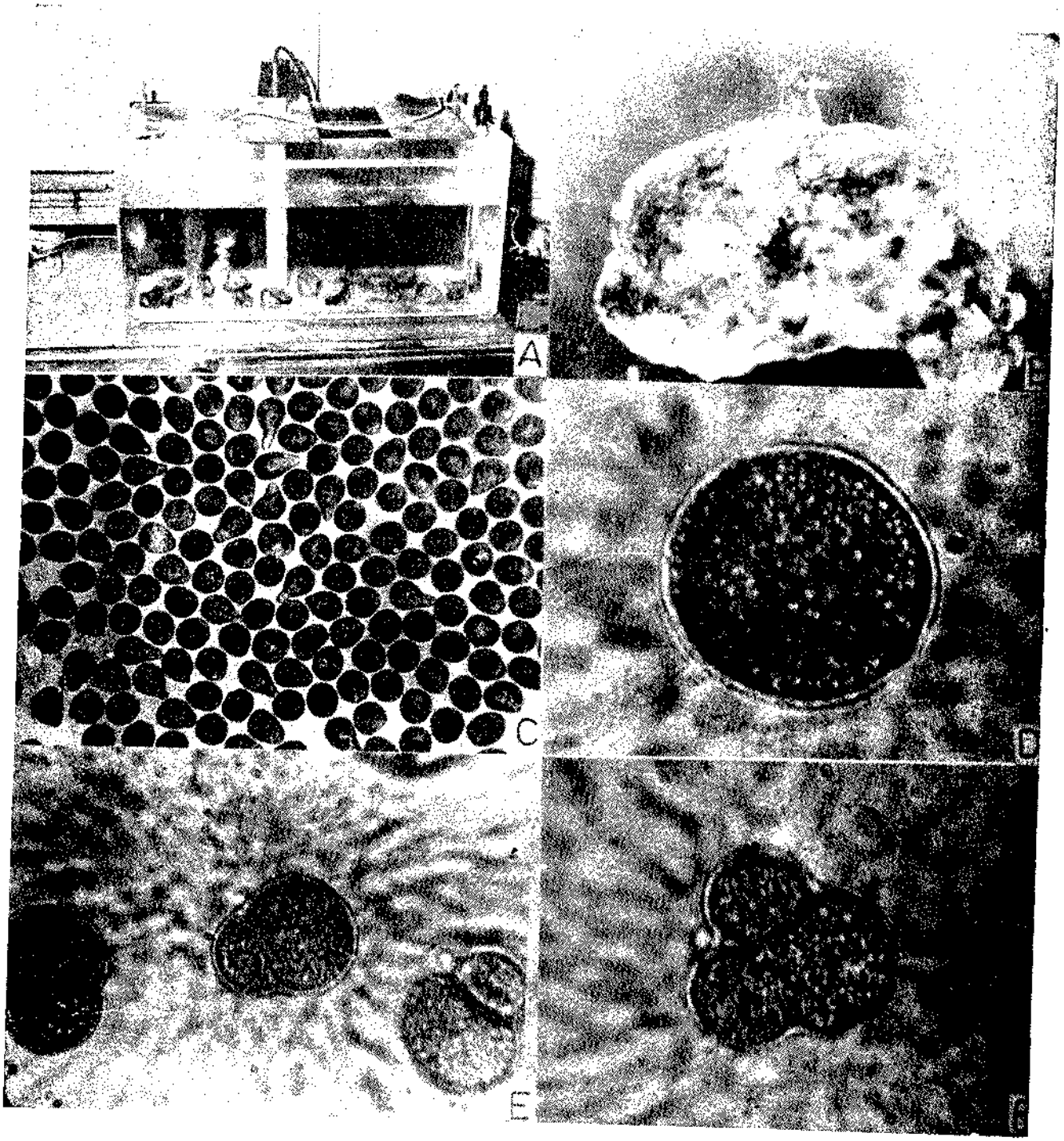

PLATE I. A. Thermal stimulation of broodstock to induce spawning in *Crassostrea madrasensis*. B. Spawning of male oyster in hatchery. C. Unfertilized eggs of oysters spawned in hatchery. D. Fertilized egg. E. Two celled stage of fertilized egg. F. Four celled stage of fertilized egg.

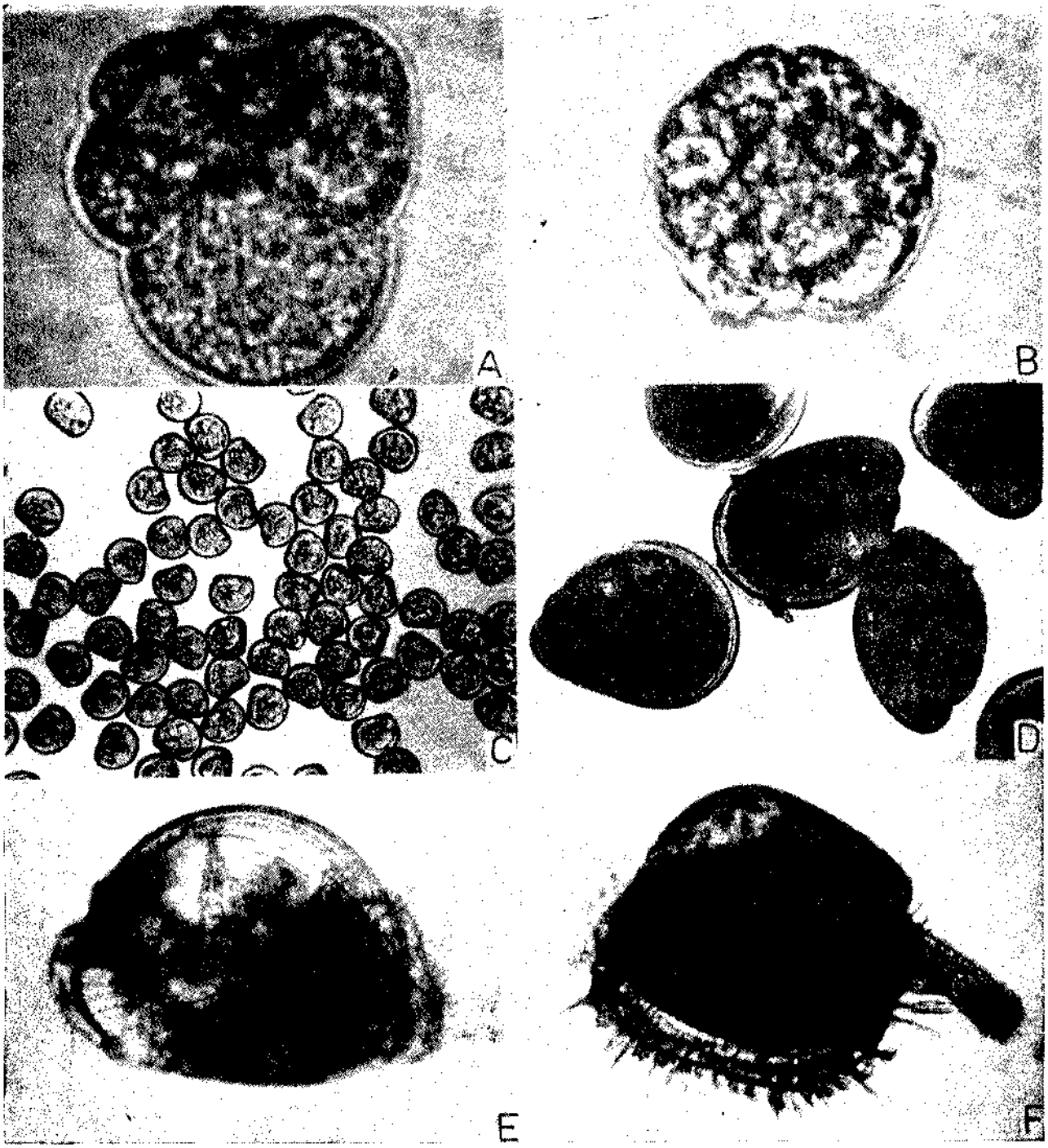

PLATE II. A. Eight celled stage of fertilized egg. B. Morula stage. C. Straight hinge stage. D. Umbo stage. E. Eyed stage. F. Pediveliger stage.

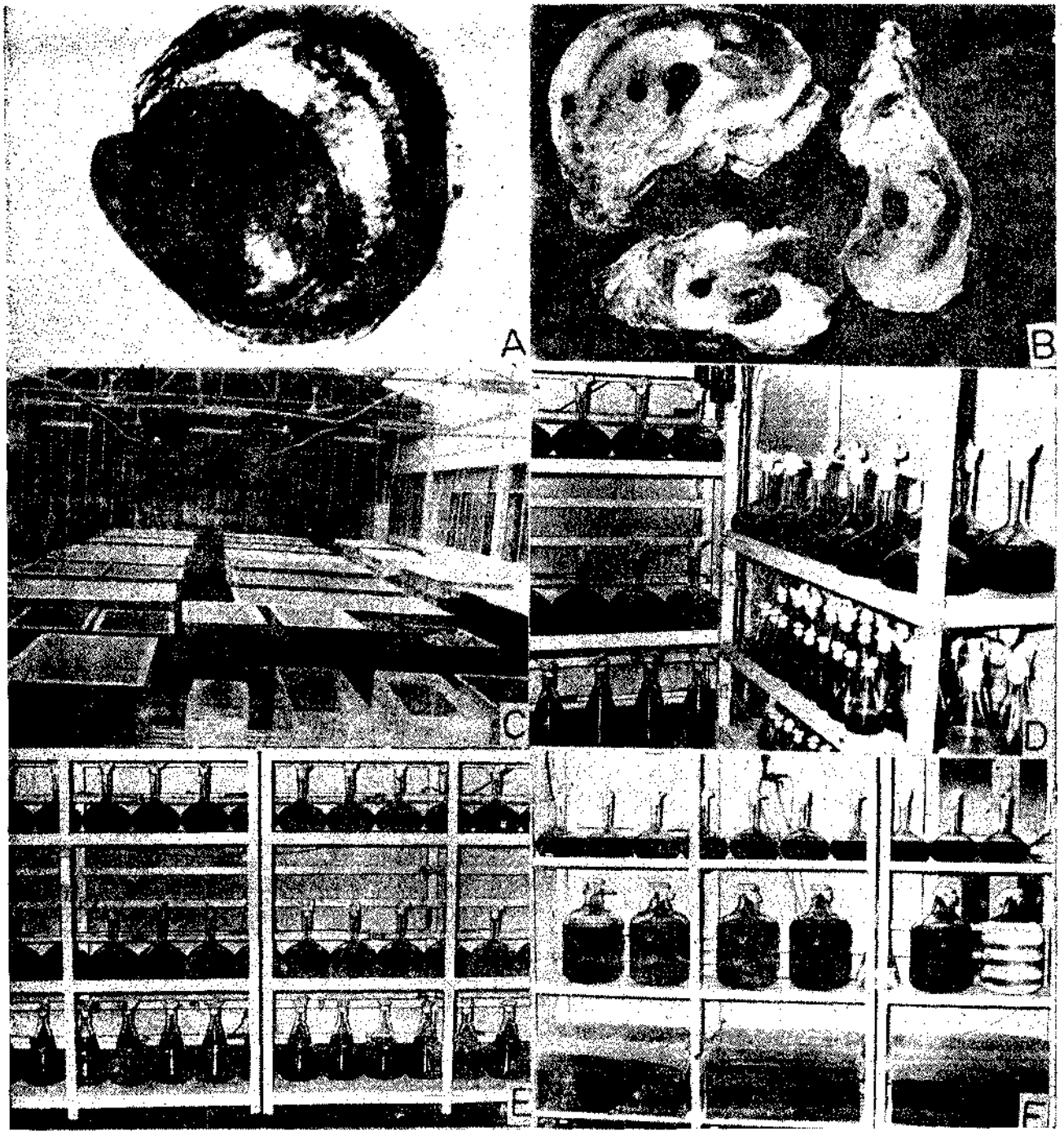

PLATE III. A. freshly set oyster spat. B. Oyster spat which had set on oyster shells. C. A view of the Oyster hatchery of CMFR Institute at Tuticorin. D. Isolation of pure phytoflagellate cultures in conical flasks in controlled conditions. E. Stock cultures of phytoflagellates, F. Mass cultures of phytoflagellates.

as to maintain the required larval density in the rearing tanks. The following densities of the larvae are advised at different stages of development during the larval period.

1. Fertilized eggs to 'D' shaped larvae : 25/ml
2. 'D' shaped stage to umbo stage : 5/ml
3. Umbo to eyed stage : 2/ml

Culture of Algal Food

The success of the hatchery operation depends mainly on the availability of adequate quantity of the larval food, the microalgae. In the natural environment, the larvae feed on nannoplankters which are readily available to them. In a hatchery the forms which will be acceptable to the larvae for their growth and development have to be identified and isolated. The production and constant supply of the algal feed, especially selected species to the larval organisms is a pre-requisite in the hatchery systems throughout the world.

Realising the importance of the nannoflagellates, measuring less than 10 microns as the essential food of the larvae of edible oyster, the isolation, identification, maintenance of stock cullure, laboratory mass culture and large scale open tank culture of these flagellates are being carried out at the molluscan hatchery of Central Marine Fisheries Research Institute at Tuticorin.

Algal species

Since the oyster larvae could feed only on nannoplankters less than 10 microns in size upto the stage of spat, suitable phytoflagellates have to be isolated from the seawater. It has been observed that the ideal phytoflagellate for feeding larvae of *Crassostrea madrasensis* is *Isochrysis galbana*, a member of the Class Haptophyceae. Apart from this, species of *Pavlova*, *Dicrateria* and *Chromulina* have also been tried as food and satisfactory results obtained. All these flagellates measure 7-8 μ and have 26-38% of protein by body weight. Once the larvae set and become spat, they are fed with mixture culture of microalgae comprising mostly diatoms and other phytoplankters.

Culture media

For the successful culturing of the microalgae various chemical culture media have been recognised depending on the organisms, Class and genera. Although most algae are photoautotrophic and can grow in purely inorganic media, many require organic compounds, the requirements of which may be either absolute or stimulatory. Usually for culturing the flagellates, Conway or Walne's medium is used in the laboratories for the maintenance of the stock culture as well as mass culture (Walne, 1970). Since this culture medium contains the chemicals, trace metals and vitamins (B_1, B_{12}) required for microalgae, the flagellates such as *Isochrysis*, *Pavlova*, *Dicrateria* and *Chromulina* are being cultured by using this media alone.

Isolation

For the isolation of the required species of phytoflagellates (Pl. III D) the serial dilution culture technique is employed. In this method mainly 5 dilution steps (the inocula corresponding to 1, 10^{-1}, 10^{-2}, 10^{-3} and 10^{-4}) or 4 steps (0.001, 0.01, 0.1 and 1 ml) are required for the isolation of the phytoflagellates. After filtering the seawater through 10 microns seive, the filtrate is inoculated to 5 series of culture tubes in various concentrations and kept under sufficient light (1 k. luxr with uniform temperature (25°C) conditions. Afte) 15 days, we can see some colouration of the culture tubes. On examination, the growth of a unialgal species could be noted in these tubes. Further purification of these organisms can be done by sub-culturing the same in 500 ml. or 1 litre conical flasks. Finally, if the culture is fully purified it is transferred into a 3 litre or 4 litre Haufkin culture flasks as stock culure.

Stock culture

Stock cultures of all the flagellates are maintained in a special room, adjacent to the mass culture laboratory. The stock cultures are kept in 3 or 4 litre Haufkin culture flasks (Pl. III E). The autoclaved or heated seawater after cooling is poured to the culture flasks and required nutrients are added. Walne's medium enriched with vitamins is the ideal one suitable to maintain the stock of all the nannoflagellates. About 10 ml of the inoculum in the growing phase is transferred to the culture flask and the latter placed in front of two tube lights (800 lux). When the maximum exponential phase has reached, only one tube light (400-500 lux) is used for further growth. Normally the flagellates will enter the stationary phase of growth after 10 days. In the stationary phase, the culture can keep for a period of 2 months in the stock culture room, under controlled conditions of light and temperature with or without aeration. At the time of maximum exponential phase of growth, the colour of the culture will turn dark brown and the cells are found in suspension without movement. It is believed that during

the stationary phase, the cells form cyst or matrix individually for thriving in the unfavourable conditions.

Mass culture

Utilizing the inoculum from the stock culture, the flagellates are grown on largescale. The containers for the mass culture of flagellates are 10 litre polythene bags, 20 litre glass carbuoys and 100 litre Perspex tanks (Pl. III F). These containers are kept in specially light provided wooden racks with aeration facilities. Fully grown culture from the stock culture room is used as inoculum for the mass culture in these containers. About 100 ml of the culture is used for mass culture in the polythene bags, 500 ml in the glass carbuoys and about 2 litres of fully grown culture is used in the 100 litre Perspex tanks providing adequate light and aeration facilities. These containers will have the maximum concentration of cells in the growing phase on 5-6th day for harvest. After noting the cell concentration using a haemocytometer, the culture is siphoned to plastic buckets or bins and supplied to the hatchery for the rearing operations. Leaving one litre of the same culture in a 20 litre glass carbuoy, fresh sterilized seawater can be added for further mass culture in the same container.

Illumination

One of the most important factors determining the successful culture of the microalgae is the type and quantum of illumination. Most of the flagellates require less light during the stationary and declining phases. Too much of light will cause the culture to decline early. For the growing phase of mass culture 1,000—1,500 lux is optimum upto 5-6 days and for maintaining the stock culture, 400-500 lux is enough for keeping the microalgae in live condition. Twelve hours of light and 12 hours darkness is ideal for maintaining the stock as well as the mass culture, which can be controlled by control switch clocks.

Temperature control

Normal temperature (28-30°C) is not ideal for the maintenance and culture of flagellates. Hence air-conditioned rooms are used for keeping the stock as well as mass cultures. Both the rooms have 23-25°C during daytime when all the tube lights are burning.

Aeration system

Similar to light and temperature control aeration of the culture tanks provides healthy culture as well as to enhance the exponential phase for a few days more. It was noticed that if aeration is given to the mass culture, the culture will remain in the growing phase for 2-3 days more than the tanks where there is no aeration. Aeration will help the nutrient salts to distribute uniformly in the medium and also for supplying carbon-di-oxide required for photosynthesis. Lastly, aeration will prevent settling out of the cells at the bottom of the culture tanks and causing eventual death due to the lack of supply of carbon-di-oxide.

Anticontamination procedure

In working with the various species of microalgae, the most important is the cleanliness of all surfaces, containers and especially of the personnel's hands. When handling the equipments, glassware and cultures, one should wash hands after working with one species and before starting to work with another species of algae. Transfer of tube cultures is done where there is a minimal movement of air to reduce chances of contamination. Further, all the stock cultures should be checked for contamination periodically using sterilized pipettes.

Harvest of the culture

The fully grown culture is harvested during the growing phase of the phytoflagellates, after determining the cell concentration. If the culture has entered the declining or stationary phase, the metabolites will be very high and the cells are not in healthy condition. The larval organisms reared will not have the expected growth if fed with such dietary organisms.

Feeding Protocol

Since the nutritional requirement increases with the growth of larvae, a schedule of feeding has been developed with different cell concentrations depending on the age and size of larvae. The cell concentration of the larval food *Isochrysis* sp. in respect of different stages of development is as indicated below.

Stages	Cell concentration in ml/larva
'D' shape ..	3,000—4,000
Umbo ..	4,000—5,000
Late umbo ..	5,000—8,000
Eyed stage ..	8,000—10,000
Pediveliger ..	10,000—12,000

Growth of Larvae

From 'D' shell stage (day 1) to umbo stage the larvae grow from a mean size of 61 μ to 156 μ in seven days with an average growth rate of 13.6 μ per day. On the 17th day the mean size of the eyed larvae is 289.2 μ with an average growth of 13.3 μ per day. On the 19th day the pediveliger is 348.2 μ showing an average growth of 29.0 μ per day.

Environmental Conditions

The water temperature in the oyster hatchery (Pl. III C) varies from 23.5 to 27.6°C on the minimal range and between 28.2 and 32.6°C at the maximum. Although the temperature varied periodically during the course of year, each experiment has been concluded within the temperature regime of the period. There was no marked fluctuation in the salinity except during November and December. The annual average salinity varied between 34.11 and 36.32‰. The pH range of the seawater measured from 7.76 to 8.20.

Seawater source and Management

The seawater required for the hatchery is drawn from the Tuticorin Bay and filtered through a bed of sand filters. Before pumping the seawater to the rearing tanks it is further filtered through sterilized cotton. In this manner the seawater is filtered effectively up to a rating of 5 μ. The salinity, oxygen content and pH of the filtered seawater are monitored periodically (twice in a week). Water temperature is recorded daily.

Equipment and Its Care

It is very important that the pipes which carry filtered seawater from the filtration facility (filter bed and storage sump) to the larval culture and setting rooms should be kept very clean and free from contamination. These pipelines should be flushed with freshwater at least once a week. The flushing of freshwater will kill the protozoans and other organisms that might get past the last filtration stage.

The filters of nylobolt and stainless steel meshed seives, after use are dipped in warm fresh water and dried.

Equipment required for spawning and rearing operations are Perspex tanks, fibreglass tanks, glass beakers and fibreglass setting tanks. Once in two days the rearing tanks are emptied and scrubbed and washed with freshwater and filtered seawater.

Aeration tubes should be periodically changed and aquarium air stones should be washed in boiling freshwater once in a week.

Preparation of Spat Collectors

The type of spat collectors used for setting of oysters should fulfil several requirements. They should be non-toxic and be favourable for oyster spat to set. They must be compact enough to allow sufficient water circulation for the growth of spat. The spat collectors should be robust enough to withstand handling and it should be possible to separate the spat individually.

The materials used for setting of spat in oyster hatchery, are oyster shells, polythene sheets, shell grit and lime-coated tiles. Oyster larvae prefer shells which have retained the natural tanned protein. Extracts of oyster tissue enhance settlement of larvae.

The spat collectors are sterilized with chlorinated water and pretreated by soaking and repeated washing in seawater for a week so as to remove any toxic substances present. The oyster shells are brushed well and washed in seawater.

Setting of Spat

The eyed larvae are released into the setting tanks of size 2 m × 1 m × 0.5 m lined by polythene sheet inside and containing filtered seawater which is aerated. The larvae are released into the tank at the rate of 2 larvae/ml. Water change is done once in two days just before feeding. The larvae are fed with *Isochrysis* at the rate of 10,000—12,000 cells/larva. The process of setting extends for 5-6 days. The spat are reared in the tank for a period of three weeks, feeding them with mixed phytoplankters such as *Chaetoceros* sp., *Skeletonema costatum*, *Thalassiosira subtilis*, *Nitzschia* spp. etc. Average setting on polythene sheet is 4 spat/sq.cm.

Oyster shell rens with 6 to 8 shells strung on 3 mm thick synthetic rope of 1 m length numbering about 25-30 are suspended in the setting tanks which are covered inside by polythene sheet. The released larvae settle in large numbers on the shells and grow into spat (Pl. III B). When the spat grow to a size of 5-10 mm the shell rens are transferred to the oyster farm for further rearing of the seed.

Cultchless Spat

Oyster shell grit and polyethylene sheets have been used for the production of cultchless spat. Oyster shell grit 500 μ in size are washed thoroughly, sterilized in seawater with 10 ppm chlorine, washed once more in running filtered seawater and dried. The shell grit are uniformly spread at the bottom of one ton capacity setting tank and oyster larvae about to set are released into it. For the setting of spat, polyethylene sheet is spread at the bottom and along the sides of the setting

tank. Three weeks after setting, the spat are separated from the polyethylene sheet, put in cages lined by velon screen and reared on racks in the farm.

Basic Facilities Required in A Hatchery for the Production of Oyster Seed

The selection of site for the hatchery is an important aspect which will determine the technological success of the system. The site should have the following environmental features. The sea water along the coast should be free from pollutants, both domestic and industrial. Waters with a wide range of salinity will not be suitable. Seawater with salinity range of 25‰-30‰ has been found to be suitable for oysters of our coasts. Places with seawater in which the level of suspended organic and inorganic matter is too much should be avoided as the cost of operation of filtration system will be high.

A set of sedimentation tanks and sand filter bed is necessary to filter a large quantity of seawater daily for the hatchery. In this system the sea water is filtered up to 10-20 μ. The filtered sea water is taken to a storage sump and is further purified by passing through cotton filters before pumping to larval and spat rearing tanks. It would be expensive to filter large quantities of seawater using microfilters. Ultraviolet light water sterilizer in which seawater is made to flow through a series of U shaped quartz tubes kept exposed to ultraviolet lamps will be required to sterilize the seawater when bacterial and fungal load is noticed in seawater.

Pump sets are required to pump seawater from the sea to sedimentation tanks and filter bed and from the latter to the storage sump and hatchery. In pipe lines PVC gate valves and pumps are used to avoid corrosion.

A 7.5 H.P. air compressor fitted with auto start switch and oil filter and a PVC pipeline system connected with it is required to supply oil free air for aerating the tanks in which the broodstock as well as oyster larvae and spat are reared.

Fibreglass tanks of various capacities 100 lit., 300 lit. and 1 ton are needed for rearing broodstock, mixed phytoplankton cultures, oyster larvae and spat.

In the room where oysters are conditioned for maturation the temperature is controlled with air conditioners.

Mass phytoflagellate culture facility in an airconditioned room with controlled temperature of 22-25°C and aseptic conditions is essential for isolation of phytoflagellates and maintaining stock cultures and production of mass cultures of the flagellates for feeding oyster larvae.

Stainless steel test sieves of mesh size 40μ, 50μ, 75 μ, 125 μ and nylobolt filters with mesh sizes of 30 μ and 40 μ are needed for segregation and culling of eggs and larvae.

A hot air oven is required to dry glassware used in culture of phytoflagellates. Chemicals for preparing culture medium for the culture of phytoflagellates have to be procured.

The development of hatchery techniques for the mass production of oyster seed is a major breakthrough which is of great significance as it will be possible to produce and supply adequate quantities of oyster seed at any time during the year for conducting oyster culture. By adopting hatchery techniques it is possible to develop disease resistant strains of oysters with regular shape, fast growth and high meat content.

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9

TECHNOLOGY OF OYSTER FARMING

K. NAGAPPAN NAYAR

INTRODUCTION

Among Hornell's (1910, 1916, 1918, 1922) suggestions for the future fisheries developmental programmes in the country was his indication about the vast scope in the realm of oyster farming in the Madras State. He initiated some experimental farming on the model of that in France. However, his attempt remained unaccomplished in the sense that there was no oyster production of significance, nor was there any organised effort in exploiting the natural resources due to the general disinterestedness of the public in oyster meat consumption. But the advancement of science and technology over the past three decades has brought about a reversal of the attitude of the public with greater awareness to utilise the protein rich marine organisms in their dietary needs. This is partly due to the realisation that agricultural production is reaching a stage where the human needs will find it difficult to meet their future requirements on account of population explosion. It is in this context oyster culture assumes special significance as the oysters are easy to grow in farms and plenty in natural abundance to supply the seed.

The technology of oyster farming is not simply collecting young oysters and stocking them in growing waters. Like scientific agriculture farming operations, it involves scientific information to achieve assured production which in turn means purposeful assessment of the prevalence and provision of adequate environmental conditions that govern healthy survival and growth. Fortunately for us in India where oyster farming is in a nascent stage, voluminous information on oyster farming, oyster biology, reproduction, larval development, growth and the role played by environmental parameters are already available in respect of countries like Japan, U.S.A., France, U.K. and Holland. Variations in these details are to be expected from one country to another. Modifications in approach to farming might become necessary while evolving suitable techniques. Governed by these considerations, the following prerequisites will have to be satisfied in the technology of oyster farming.

(1) Resources availability.
(2) Study of the biological aspects, growth and reproductive cycle.
(3) Study of the environment : hydrological parameters and plankton food availability.
(4) Selection of suitable farm site.
(5) Experimentation in spat collection and standardization of techniques.
(6) Growing technique and standardization of methods which are suitable.
(7) Establishment of model farms and their management. (Fabrication of culture materials, predator and fouler control, avoidance of disease factors, labour and material management).
(8) Harvesting, post-harvest strategy, marketing and product development.
(9) Socio-economic feasibility and cost effectiveness.
(10) Extension and training.

Keeping these above in view, efforts to evolve the technology for oyster farming were started by Central Marine Fisheries Research Institute at Tuticorin which have yielded satisfactory results. The techniques have been repeatedly tested over a period of five years and found to give consistent results at the same time opening up possibilities for trying out alternate cheap and more effective methods. The standard techniques adopted

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001,

are the first of their kind in our country and it is hoped to serve to inspire development of techniques in respect of other cultivable edible molluscs of our coastline.

Culture Methods

Two important aspects are involved in oyster culture i.e., production of seed and rearing of the seed oysters to marketable size. Nayar (1980) has given the description of the various methods i.e., raft method, rack and tray, stake method, long-line method and on bottom method by which oysters are cultured. In the on-bottom method, oyster seed are sown and allowed to grow on bottom in shallow coastal areas. The production is low but because of economic considerations it is still being followed in U.S.A. In the stake method, wooden stakes are planted in coastal waters and oyster spat set on these and are allowed to grow. In rack culture, oysters in rearing trays are placed on a platform constructed with wooden poles, in shallow and calm areas with depth ranging from 1-1½ metres. If the depth is more, oysters in strings can be reared by suspending from a raft. The long-line method is a modified form of the raft method. The long line unit consists of a series of wooden or styrofoam floats, to which two parallel synthetic ropes of 6 cm thickness are tied. From these paired horizontal ropes, rens are suspended and the length of the rens depends on the depth at the place. It was decided that the rack culture technique is suitable at Tuticorin because of the shallow nature of the bay where large scale operations of oyster culture could be carried out.

Location of farm

The oyster farm is situated in Tuticorin Bay on the southeast coast of India Lat. 48°N, Long. 78°11'E. The racks are erected in the bay, in the tidal zone where the water depth varies from 0.5 m to 1.5 m. Salinity of the bay ranges from 29.4 to 35.3‰. Rarely in monsoon season, due to heavy rainfall and discharge of freshwater from creeks in the area, the salinity may drop to 15‰. The temperature ranges from 25°C to 31°C. The water temperature is high during April-May which is the peak spawning season of *Crassostrea madrasensis*. During January-February, the water temperature is 25°-28°C.

Rack

Six vertical poles of 2.4 m length are driven to the bottom at an interval of 2 m apart and another set of six poles are driven parallel to the first row. These two rows of poles are connected by 2 m long cross poles. Above these cross poles, using 8 poles 5.5-6.5 m in length a platform for keeping oyster rearing trays is constructed. Coir and 3 mm synthetic ropes are used as the binding materials. Each rack occupies an area of 25 sq.m. and accommodates 20 rectangular trays with holding capacity of 3,000-4,000 oysters.

Seed collection

Eventhough various types of spat collectors were tested for their suitability in collection of oyster spat, for large scale collection, materials such as lime coated tiles and oyster shell rens were used. Thangavelu and Sundaram (1983) have given an account of lime coated tiles of size 24 × 15 cm used in spat collection. The lime coated tiles are arranged in trays at the rate of 50/tray in such a way that the concave side of the tiles faces downwards.

Oyster shell strings are prepared from 20-25 number of oyster shells centrally punctured and strung on 1½ metre long G.I. wire. Each unit is known as a 'Ren' and about 95-100 rens are placed horizontally on a rack for spat collection. The aspects of laying spat collectors, the number of spat per different spat collectors, the number of spat collected during different years are dealt in a separate chapter.

Rearing of seed oysters

The method of rearing seed oysters depends upon the type of spat collectors used. Those which had set on oyster shells could be allowed to grow upto marketable size on the spat collectors themselves. Fresh rens have to be prepared with the spat set shells giving sufficient interspaces to enable growth of oysters and the rens are hung from the racks. The shells with spat can be removed from the strings and individually they can be used for an on bottom culture in suitable shallow areas.

Spat set on limecoated tiles are allowed to grow on the tiles for a period of two months. When the spat attain a size of 25 mm, the layer of lime with spat it scraped off using a scraper. The scraped tiles are reutilised for spat collection, after giving a lime coating. The detached spat are reared in box-type cages of size 40 × 40 × 10 cm made of 6 mm. M.S. round rod knitted with 2 mm synthetic twine. The cages with oysterlings (150-200 numbers/cage) are suspended from racks using 4 mm thick and 1½ m long synthetic ropes. For suspending the box-type cages or the rens prepared with spat set shells, racks are constructed by erecting six vertical poles (2.4 m in length) at a distance

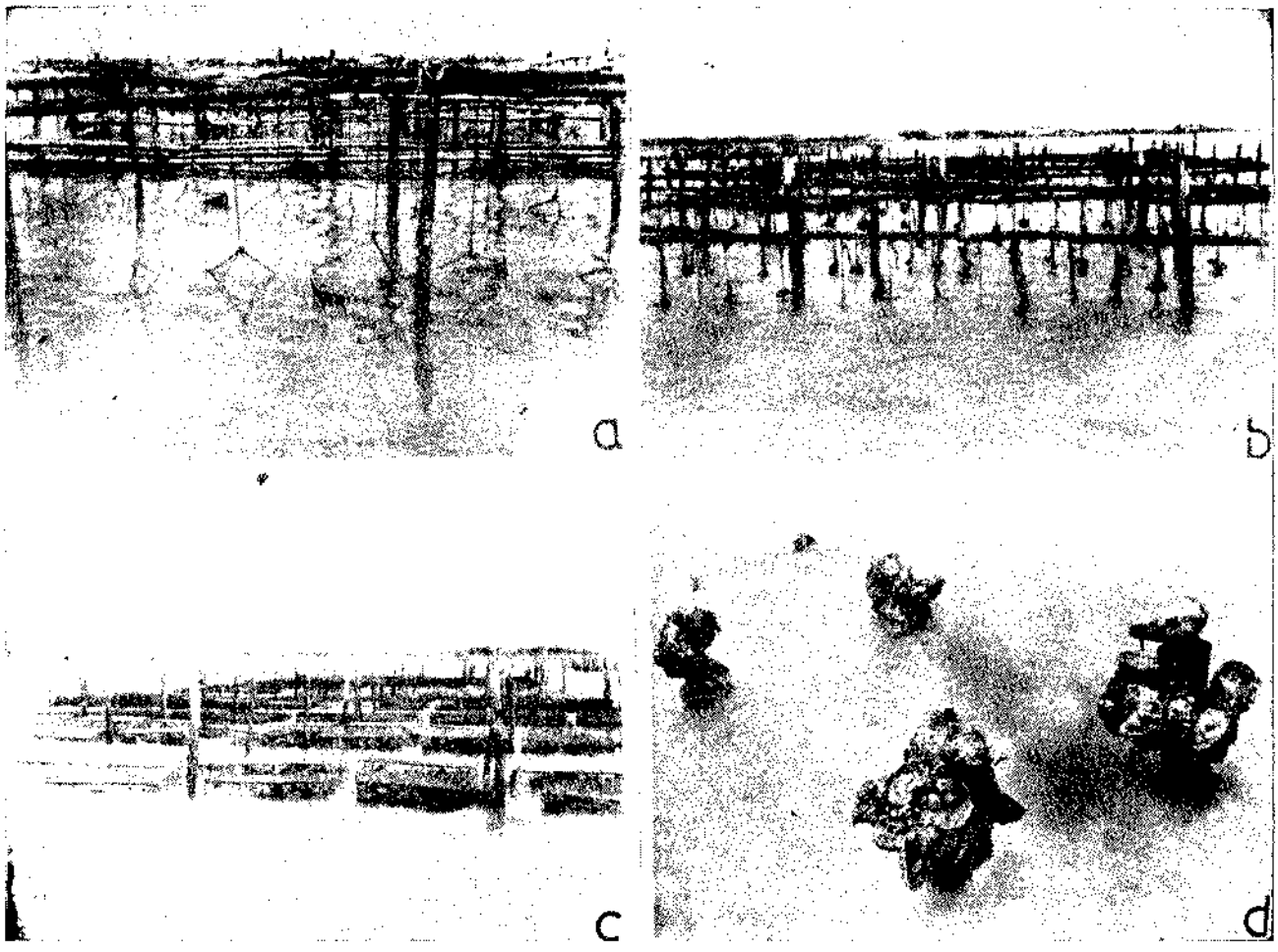

Plate I. (*a*) Box-type cages suspended from racks, (*b*) Strings hung from racks, (*c*) A view of oyster farm with racks on which trays containing oysters are kept, (*d*) Oysters grown on stakes.

of 2 metres. Long horizontal poles are tied across these poles at the top at a height of 1.8 m. From the horizontal poles, the cages (Pl. I a) or strings (Plate I b) are suspended.

Thinning and growing oysters

Oysters which have grown to 50 mm size and above are segregated from the box type cages and transferred to rectangular trays of size 90 × 60 × 15 cm each tray holding 150-200 oysters. Twenty such trays are kept on a rack (Pl. I c). The oysters are reared from settlement, for a period of one year when they attain marketable size of 80-90 mm, weighing 80-100 gm with meat forming 8-10%. During rearing, periodical cleaning of oysters, cages and trays and maintenance of racks have to be carried out for healthy growth of oysters.

With successful mass production of oyster seed in the CMFRI oyster hatchery at Tuticorin, the spat which had set on shells in the hatchery are being utilised for stake method of culture (Pl. I d).

Harvesting and marketing

The meat of oysters in sexually ripe stage are creamy in colour, tasty and in best condition with maximum weight and the oysters have to be harvested when they are in that stage. Soon after spawning, the oyster meat will be thin and not very tasty. The condition factor of the oysters ranges from 40 to 180. The higher condition factor is found before the spawning season i.e. April-May and August-September. When the meat is plumpy and creamy about 100-120 oysters yield one kg of meat. Harvesting of oysters is done manually. Harvested oysters are cleaned with a jet of water and purified by keeping them in filtered, unpolluted sea water for 12-15 hrs. The oyster meat is shucked after placing the depurated oysters in hot water for 2 minutes. After washing thoroughly, the shucked meat is sold locally. On three occasions, large quantities of cultured oysters were harvested and the shucked meat was sold to Integrated Fisheries Project, Cochin. The meat was quick frozen, transported in insulated van and are either smoked and canned or canned in brine. It was sent to several far off places for consumption (Samuel *et al.*, 1982).

Remarks

Oyster culture at Tuticorin (Nayar and Mahadevan, 1983) and some of the experimental culture carried out in Athankarai estuary (Rao, *et al.*, 1983) Bheemunipatnam backwaters, Andhra Pradesh (Ruben *et al.*, 1983), Mulki estuary (Mohan Joseph and Shantha Joseph, 1983), Goa (Parulekar *et al.*, 1983) and Cochin backwaters (Purushan *et al.*, 1983) indicate good prospects for oyster farming along the Indian coasts.

The rack culture technique developed at Tuticorin could be profitably employed to conduct oyster farming in the stretches of shallow coastal waters at several places along the Indian coasts. Bottom sowing method of culture should also be attempted in shallow areas with hard bottom in view of the advantage of low investment needed.

Adverse conditions of hydrographic or biological factors such as predation, parasitism and disease may cause considerable damage to the tended stock. Though at present pollution is not posing a big problem, in the near future, because of increasing industrialization along the coastal areas, it may cause environmental problems. Proper treatment of domestic, oil and thermal pollutants will reduce the injurious effects on the environmental conditions.

Predatory gastropods cause mortality of oysters when they are reared in box-type cages (Thangavelu and Muthiah, 1983). Rao *et al.* (1983) have mentioned the predation of cultured oysters by the crabs *Scylla serrata* and *Pagurus* spp. in Athankarai estuary. *Balanus amphitrite* is a serious one among the pests in tropics and competes with oysters for space and food. The wood borers *Teredo furcifera, Lyrodus pedicellatus, L. affinis* and *Martesia striata* cause damage to the poles with which oyster culture racks are constructed (Nair and Dharmaraj, 1979). Studies on the remedial measures for these problems have to be carried out to facilitiate better production from oyster farming.

The meat of oysters cultured at Tuticorin is free from pathogenic bacteria. Heavy metal contaminants have been found to be well below the admissible limits. But the recent incidence of paralytic shellfish poisoning in Vayalur village, Tamil Nadu (Silas *et al.*, 1982) and in Kumble estuary near Mangalore (Karunasagar, 1984) indicate the need for monitoring studies on the sanitary quality of shellfish growing waters, shellfish toxicity and occurrence of toxic dinoflagellate blooms in the areas where bivalves are cultured. Depuration of oysters before marketing them, should be made mandatory.

Canning and marketing of oysters cultured at Tuticorin has shown that there is good demand for canned oysters. More extension work is needed for disseminating information on the food value of oysters, techniques of oyster culture and the various uses of

oyster shells i.e. in calcium carbide and cement industries and poultry farming apart from its use in the production of lime which could lead to adoption of oyster culture practices commercially.

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10

POST-HARVEST TECHNOLOGY

M. E. RAJAPANDIAN[1] AND P. MUTHIAH[1]

INTRODUCTION

In recent years great interest is envinced in several countries in the culture of molluscan shellfish. In the tropics favourable conditions exist for rapid growth and marketable oysters are obtained within ten to twelve months. Investigations have been conducted at the Central Marine Fisheries Research Institute and success has been achieved in perfecting the techniques of oyster culture in coastal waters (Nayar and Mahadevan, 1983). The awareness that shellfish have to be purified and rendered harmless goes back to the time of the Roman Empire. During the first century B.C. the Romans consumed cockles and oysters after treating them in tanks known as cockle washery (Yonge, 1962). Presently the commercial producers of shellfish in many countries, usually follow purification procedures though they differ from country to country. While chlorinated seawater has been widely in use for depuration of molluscan shellfish, recently U.V. or ozone treated seawater has come to be used in French and Australian shellfish depuration plants. Harvesting of oysters is done during prespawning period, when the meat condition is good. The spawning season of *Crassostrea madrasensis* is April-May with a mild secondary spawning period in August-September. High condition factor of 90-150 has been observed before spawning indicating the plumpness of the meat. After spawning the meat is watery and thin. Harvesting method of oysters differs according to the method of culture. Dredging, tonging and handpicking are some of the harvesting methods for oysters which are cultured on bottom. At Tuticorin, since oysters are cultured in trays, harvesting is done manually and the oysters are transported to depuration plant in a fibreglass reinforced plastic dinghy (Pl. I a).

In this chapter the techniques followed in India in handling and processing oysters after harvest are described.

PURIFICATION

Oysters being filter feeders may harbour bacteria and other microorganisms and may also accumulate contaminants present in the surrounding water which is known as bioconcentration. The reverse process called depuration is the process of purification by which the shellfish are rendered free of the harmful materials. In depuration, the shellfish are placed in tanks supplied with seawater artificially sterilized. The process may either be continuous or discontinuous. In continuous depuration system, 10-20% seawater in the purification tanks is continuously renewed with running filtered seawater. In the discontinuous system the frequency of the water change ranges from two to three times a day (Fauvel and Pons, 1978).

Nayar *et al.* (1983) have designed and operated a simple method to purify the oysters cultured in the Institute's farm at Tuticorin. This method assures effective purification of oysters at the rate of 14,400 oysters (1,300 kg) per day. The oysters are arranged in twentysix synthetic twine knitted trays each measuring 60 cm × 60 cm × 15 cm. and placed on wooden frames which are arranged in a concrete tank measuring 2.5 m × 2.5 m × 1.0 m. Firstly the oysters are thoroughly hosed by a strong jet of filtered seawater (Pl. I b) to remove external mud and dirt which are flushed out through the outlet of the tank. By this time, the purification tanks each measuring 2.5 m × 2.5 m × 1.0 m. are got ready and filled with seawater to a height of 70 cm. In each tank, trays with oysters

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001.

numbering 2,400 are placed. A continuous, slow flow of seawater into the tanks and exit through the drain valve is kept up so as to maintain a water column of 70 cm inside the tank. The oysters are allowed to remain in the tank for twelve hours to rid the oysters of microorganisms.

Subsequently seawater in the purification tank is drained and the oysters are once again hosed with a strong jet of seawater. By this procedure the accumulated faeces are removed from the tank. The purification tank is again filled with filtered seawater, the oysters are relaid and the process is repeated for another twelve hours. At the end of this period, the oysters are kept for one hour in seawater purified by treating with 3 ppm. chlorine. Then the oysters are washed once more in filtered seawater and kept ready for marketing. This procedure makes the oysters free from bacteria and renders them suitable for human consumption.

Transport and Storage of Live Oysters

Oysters can survive out of water for several days, if carefully handled and kept moist and cool. However, it is desirable that the oysters reach the consumer market within three days of harvest if they are to be in prime condition. They should be transported in wet gunny bags which are kept moist from time to time. They should be kept in a manner that protects them from mechanical damage. The method of packing depends on the value of the product, journey time and the market for which it is sent (Stroud, 1980).

For marketing purpose, consignments of 300 oysters each kept in wet gunny bags and packed in bamboo baskets were sent twice by train to Madras 560 km away. The consignment reached the destination in a wholesome condition with 'nil' mortality. For processing studies, 5,500 oysters packed in 37 wet gunny bags were transported by road for twelve hours to Cochin. There was no mortality either enroute or at destination. This indicates that live marketable size oysters packed in wet gunny bags can be safely transported for 25-30 hrs, without mortality. Small holding tanks having filtered seawater or artificial seawater and provided with adequate aeration facilities would keep the oysters alive for a few days at the wholesalers premises.

Shucking

Shucking is the removal of meat from the oyster. One develops skill in shucking after some practice. Further each person develops his own individual opening technique.

The materials required for shucking are a shucking table, shucking knives, a perforated stainless steel table and rubber gloves for preventing cuts from the sharp edges of the oyster shell. The shucking knife is made of stainless steel, 30 cm long, has a stout wooden handle and the distal end is flattened into a cutting edge of 2.5 to 3.0 cm (Quayle, 1969).

For shucking the oyster it is placed on the table with the cupped or left valve down with the hinge pointed towards the opener's left. Sometimes experienced shuckers hold the oyster in this position in their left hand and do the shucking (Pl. I c and d). The sharp edge of the shucking knife is inserted between the two valves of the shell close to the hinge (Fig 1). After the knife edge has entered inside, the blade is forced

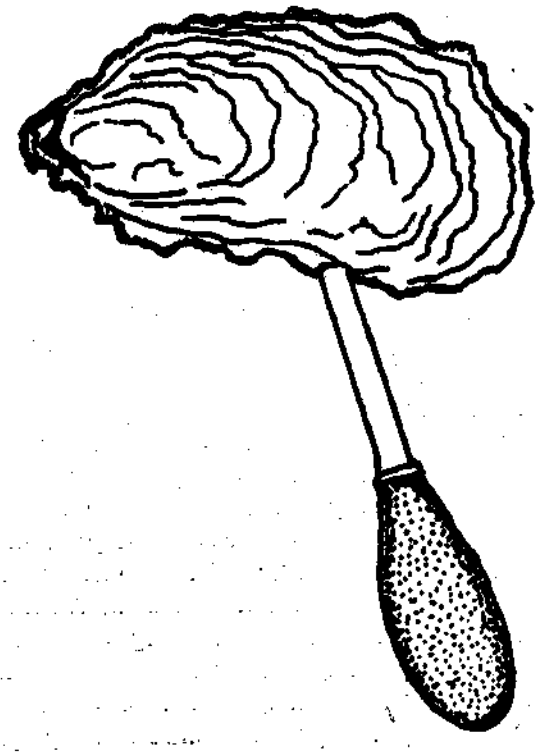

Fig 1. Position of shucking knife in the oyster while opening.

further into the shell cavity of the oyster to about 2 to 3 cm when a movement of the knife to the right and left will sever the adductor muscle of the oyster. A prying motion of the knife will break hold of the hinge and the two shell valves of the oyster will separate. The upper valve is discarded, the oyster meat is separated by cutting the base of the adductor muscle attached to lower valve and flipped into the container. If the oyster is to be served on the half shell, it is cleaned of any shell fragments and left in the cupped lower valve.

Plate I. *a* Harvested oysters. *b* Oysters kept in tank for initial cleaning. *c* and *d* Oyster meat being shucked from the depurated oysters.

Several methods have been tried to reduce the labour of hand shucking. These include the shearing of the hinge or beak of the oysters by guillotine and a wide range of treatments that cause the shell to gape open, including the use of chemicals (weak hydrochloric acid), heat, cold, vacuam, microwaves and lasers (Wood, 1972). Among all these methods, the easiest is freezing the oysters before shucking (Stroud, 1980) or placing the depurated oysters in a hot-water tank in which temperature is not high enough to cook the oysters, but sufficient to open them (Nowak, 1970). A skilled shucker can remove meat from 120 to 150 oysters in an hour.

Processing of Oyster Meat

Freezing :

After purification whole oysters (shell on) can be frozen satisfactorily spread in single layers in an air blast freezer. Oysters can also be frozen in the half-shell. They should be laid in a single layer on trays in an air blast freezer with polypropylene film stretched over each tray to protect the open surfaces of the oyster meat.

Frozen whole oysters packed in polythene bags can be kept in good condition for six months in a cold storage at —30°C. The liquid within the shell acts as a glaze to protect the meat from dehydration. The meat of frozen whole oysters is suitable for preparing dishes.

Freezing Oyster Meat

If the demand for frozen whole oysters is not much, it is economical to freeze shucked meat. Oyster meat frozen either individually or in blocks (in 1 kg or 2 kg slabs) will yield an excellent product after thawing.

Canning of Oyster Meat

Oyster in brine :

Oyster meat is chilled, washed and blanched in 3% brine containing 0.1% citric acid for 4-5 minutes. Ninety gm of blanched meat is packed in cans (112 gm net wt.) and a hot 2% brine with 0.1% citric acid is added. The packed cans are seamed and sterilized at 115°C for twentyfive minutes. The cans are then chilled immediately, wiped to remove water present on the outside and kept in storage (Stroud, 1980).

Smoked Oyster :

For preparation of smoked oyster meat, the meat in washed, treated with 5% brine for 5 minutes, drained, dipped in edible oil, spread in a single layer on nylon wire mesh, drained again and loaded into the smoking chamber (Samuel *et al.*, 1982). The meat is held is dense smoke and maintained at a temperature of 40°C for 30 minutes and later at 70°C for 80-90 minutes. In the smoking chamber the materials are turned over once in 15 minutes to ensure uniform smoking of the meat. The smoked oysters are packed in cans and sufficient quantities of hot refined edible oil is added. The cans are then seamed, sterilized at 115°C for 25 minutes and immediately cooled and stored for marketing (Stroud, 1980).

Meat of cultured oysters harvested from oyster farm at Tuticorin was shucked and processed. The shucked meat was washed well and dipped in 1% salt solution containing 0.2% citric acid to avoid drip loss. The meat was then packed in 2 kg units, quick frozen and transported at—20°C in insulated van to Integrated Fisheries Project, Cochin. The meat thus transported was canned either in oil or as smoked oysters. About 2,218 cans with net weight of 80 gm of oyster meat were obtained from 500 kg of oyster meat (Samuel *et al*, 1982) at the cannery of the Integrated Fisheries Project, Cochin. The canned oysters were sold in major cities of India and was well received.

Utilization of Oyster Shells

The oyster shells constitute about 90% of the total weight and contain 52-55% of calcium oxide and therefore are suitable raw material in calcium carbide, lime, fertilizer, cement and other industries. Further, the oyster shells are ideal for use as spat collectors for the collection of oyster spat. The shells can also be distintegrated to suitable size and used as poultry grit.

Oyster shells (100 kg) were disintegrated and shell grit of four different grades 5 mm, 2 mm, 1 mm and $\leq$ 1 mm were obtained. On trying them in a local poultry farm 2-3 mm size grit was found to be suitable as an ingredient in poultry feed. The bulk of the oyster shells were sold to calcium carbide industry since they could be disposed of without incurring any expenditure towards processing as in the case of shell grit. The shells are also being utilized as spat collectors in the hatchery as well as in spat collection from nature.

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11

ECONOMICS OF OYSTER CULTURE

K. NAGAPPAN NAYAR[1], S. MAHADEVAN[1] AND P. MUTHIAH[1]

INTRODUCTION

Economic analysis of any system of aquaculture practice aids not only to improvise management practices but also ensures profitability. Attention on return on investment has been rightly emphasized by Mitchell and Usry (1967), Pillay (1973) and IPFC (1975) to show that well planned and properly managed aquaculture ventures compare very favourably with similar other food production industries. Hornell (1910) realising the edibility of the oyster meat and its nutritional value initiated efforts on oyster farming at Pulicat Lake and gave an approximate account of working expenses of a one ha. park. These estimates are not relevant to the present day cost but nevertheless provide an idea of the material inputs that have to go into the system. Blanco and Montalban (1955) have worked out the economics for one ha. oyster farm. Quayle (1971) and Humphries (1976) have given the production cost of oysters cultured by raft method and economics of tray culture respectively. Similarly Blanco (1972) has given the investment returns for oyster farms in Philippines. Koganezawa (1979) has stated that it is difficult to arrive at the production cost of oysters in Japan due to the wide range of culture methods and efficiency. Moreover, these enterprises are owner-operated. It is thus clear that for aquaculture, to become important in national economy the cost effectiveness is vital and it should be technologically practical and also fit into the legal and economic structure (Hanson, 1974). The technology of oyster farming experiments conducted at Tuticorin by 'rack' method has been explained by Mahadevan *et al.* (1980) and Nayar and Mahadevan (1983). Following this it was felt necessary to explain the economics of this system of oyster farming.

[1] *Present address :* CMFRI, Research Centre, Tuticorin-628 001.

THE FARM AND YIELD

Provision for an oyster farm with 90 racks for growing a stock of 500,000 oysters was made in the intertidal region of the Tuticorin Bay. Of this, a unit of 60 racks covering an area of 0.25 ha. was taken for model analysis. Each rack covered an area of 25 sq. m. with 20 trays accommodating, 4,000 oysters. The actual yield of oyster meat from this was 2,475 kg which works out to 9% of the total harvested stock.

COST STRUCTURE

The economic evaluation of 0.25 ha. oyster farm was calculated on this basis and presented in Table 1.

I. A. Initial investment :

(*a*) *Dinghy :* A fibreglass dinghy at a cost of Rs. 7,000 was used in transportation of farm materials to and from shore and farm area. The cost could be amortised over five years at Rs. 1,400 per year. If carefully handled the dinghy will be good for more number of years.

(*b*) *Rack :* For construction of a rack, 17 teak poles of 5-6 m length and 6-8 cm diameter were required. The cost of teak poles, tar coating, binding materials (coir and synthetic i.e. polypropylene ropes) and construction charges together amounted to Rs. 250 per rack. Each rack would serve for three years. Amortising the total cost for 60 racks (Rs. 15,000) yearly the annual cost worked out to Rs. 5,000.

(*c*) *Rearing trays :* 1,200 rearing trays of size 90 × 60 × 15 cm with synthetic webbing were good for three years, and at an initial cost of Rs. 40 per tray, the annual cost would be Rs. 16,000.

(*d*) *Box-type cages :* For initial rearing of spat, box-type cages of size 40 cm × 40 cm × 10 cm were used. The cost per tray was Rs. 25 with three years durability. The annual cost towards 800 cages would be Rs. 6,667. The rearing trays and cages together formed major portion (74.9%) of the annual capital cost.

(*e*) *Synthetic rope :* Synthetic rope used to suspend the box-type cages from the racks cost Rs. 833 annually.

II. Operational cost (O.C.) :

(*a*) *Seed :* For stocking the 1,200 trays in 60 racks, total seed required was 3 lakhs. The cost towards collecting the seed came to Rs. 6,000 accounting for 27.3% of operational cost.

(*b*) *Maintenance and repair :* Replacement and repair of racks and trays, forming 13.6% of the O.C. had cost Rs. 3,000.

Table 1. *Economic evaluation of oyster culture by rack method in* 0.25 *ha.*

Sl.No.	Items	Quantity	Initial cost Rs.	Annual depreciation Rs.	%
I. *A. Initial investment :*					
(*a*) Dinghy	..	1	7,000.00	1,400.00	4.6
(*b*) Racks	..	60	15,000.00	5,000.00	16.5
(*c*) Rearing trays	..	1,200	48,000.00	16,000.00	52.9
(*d*) Box-type cages	..	800	20,000.00	6,666.66	22.0
(*e*) Synthetic rope	..		2,500.00	833.33	2.8
(*f*) Farm accessaries	..		1,000.00	333.33	1.1
Total			93,500.00	30,233.32 or 30,233.00	99.9
I. B. Interest @ 12%	..		11,220.00	11,220.00	
			1,04,720.00	41,453.32	
II. *Operational Cost* :					
(*a*) Seed 3 lakhs	..			6,000.00	27.3
(*b*) Maintenance and repair	..			3,000.00	13.6
(*c*) Labour	..			8,000.00	36.3
(*d*) Predator eradication	..			2,000.00	9.0
(*e*) Harvesting	..			3,000.00	13.6
Total	..			22,000.00	99.8
III. Annual cost of racks and trays operational cost and			..	63,453.00	
IV. Gross income through sale of oysters @ Rs. 37/kg. for 2,475 kg of meat			..	91,575.00	
V. Net income			..	28,122.00	
Ratio to annual cost	..	44.3			
Ratio to Gross income	..	30.7			
Ratio to investment	..	30.1			

(*f*) *Farm accessories :* Farm accessories i.e., iron stand, hammers and scrappers etc. cost annually Rs. 333 forming 1.1% of the fixed cost.

Thus the initial cost came to Rs. 93,500 and the annual cost worked to Rs. 30,233. The cost towards the purification of oysters, salary of supervisory staff and rent of land were not included.

I. B. Interest :

Interst at the rate of 12% on investment worked out to Rs. 11,220.

(*c*) *Labour cost :* The annual labour cost incurred towards maintenance of rack and periodical cleaning of oyster cages was Rs. 8,000. This formed 36.3% of the O.C. During July to November when predation of stock by the gastropod *Cymatium cingulatum* was intensive, labour cost involved in eradication came to Rs. 2,000 forming 9% of O.C. Harvesting entire stock and subsequent cleaning cost came to Rs. 3,000. Thus the total labour cost formed 72.5% of annual O.C.

Ratio to Investment

The total annual production cost towards 2,475 kg of oyster meat came to Rs. 63,453. The objectives of oyster culture at Tuticorin were two-fold. One is to develop a suitable technology of farming oysters to marketable size and the other is to popularise oyster culture and establish a market for the oysters produced. Since this is altogether a new product for consumption by local people, steps were taken to distribute samples of shucked meat to public and different agencies to ascertain their opinion about the quality and palatability of the oyster meat. Major portion of harvested oyster meat was utilised for this extension work and also towards evolving suitable processing technology like smoking, deep freezing and canning. In an effort to popularise the oyster meat consumption amongst the public, a basic market price of Rs. 16 was arrived at although Rs. 37 per kg could be the actual worked out cost considering expenditure and inputs. While calculating the economics, the latter has been taken as the criterion. In order to popularise, oyster meat was sold locally at Rs. 16 per kg. Realising that the price could go up with popular demand in future years a price of Rs. 37 per kg of oyster meat was taken for calculating the ratio to investment. The sale of oyster meat at the rate of Rs. 37 would fetch Rs. 91,575. The net income could be Rs. 28,121. The break-even price of one kg of meat produced was Rs. 25.63. The break-even proudction would be 1,714 kg of oyster meat.

Ratio of net income to the annual cost works out to 44.3%. The ratio of net income to investment is 30.1% which is better than the return furnished by Blanco (1972) for Bacoor Bay oyster farm (20.5%) and slightly lower than the return from Binakayan Demonstration Farm, Philippines (38.48%).

Remarks

Gerhardsen (1979) remarks that the natural resources, labour and capital are the main factors affecting production from aquaculture. Regarding natural resources Hornell (1910, 1914, 1917, 1922), Rai (1928), Rao (1963), Alagarswami and Narasimham (1973) and Nayar and Mahadevan (1974) have drawn attention to the potentialities and availability of edible oysters all along the Indian coasts, particularly the east coast. Nayar and Mahadevan (1983) estimated that at least a considerable portion of 2 million ha. of backwaters and brackish waters area could be profitabily utilised for oyster culture along the east coast. For the present the requirement of seed for the above farming activities, can be easily procured from the intertidal open sea bay area following the 'tile' collection technique. It is fully recognised that seed collection dependant on natural resources has to be eliminated ultimately by developing a seed production technique through hatchery system. The Central Marine Fisheries Research Institute has taken note of this priority area for research and development devoting attention to achieve a breakthrough in hatchery production. Successful hatchery production of oyster spat has been achieved and the Tuticorin hatchery is at present in a position to ensure production of any number of oyster spat needed for large scale culture (Nayar *et al.*, 1984).

The aim of entrepreneurs will be to maximise income or to maximise return on investment. The return can be maximised by mechanisation at any desired level as Korringa (1976) observed that the return from mussel culture outgrew Dutch oyster industry because of mechanisation. Mechanisation can effectively reduce the labour cost which forms 72.5% of the operational cost.

Maximisation of income can also be achieved by effective prevention of predation since eradication of predators cost 9% of operational cost. Mackenzie (1970) calculated that $ 40 spent for chemicals used in eradication of predatory drills, increases the production cost of oysters. This is an area needing further research.

Production cost of oyster differs according to the culture method adopted and the scale of operation. Rabanal and Shang (1979) stated that the economic profitability of aquaculture can be improved, not only by increasing productivity, but also by reducing the production cost, and improving prices of the product. The production of rack and tray system can be increased two fold by adopting two tier system of arranging the trays one below the other unlike the present single file system (Nayar and Mahadevan, 1983). There is ample scope to reduce the cost by devising cheaper and suitable cages instead of the present synthetic netted iron frame trays and cages which cost 74.9% of the annual capital cost. A series of successful oyster culture experiments conducted here in 1985 using rens indicate the bright prospects of bringing down the cost on initial investment in regard to items (c) and (d) and on items (a) and (b) under O.C.

Preparation of the harvested oysters prior to sale in the market adds expenditure to the total cost. Especially in the culture of molluscs, the cost incurred on purification is an additional expense. If aquaculture is organised on co-operative basis, the purification system can be developed as a common facility thus reducing capital cost.

Towards obtaining high returns for the product, local demand for oysters has to be created by evolving better marketing strategy and creating market channels. Realising the potentiality of consumer demand from distant interior places and the market in foreign countries, steps for properly canning the oyster meat have been taken up by CMFRI in collaboration with Integrated Fisheries Project, Cochin.

Mariculture practices have to be classified as high-risk activities due to the enigmatic variations in the environmental parameters and their adverse impact on the culture operations by natural calamities, diseases, predation and pollution due to agricultural and industrial effluents. Crop insurance cover is one of the means for mitigating the possible losses. Although agricultural operations have been protected now by insurance cover it would take time for recognition in mariculture. In the meantime some suitable methods for evolving reasonable premia levels for aquaculture stocks can be formulated.

By blending sea farming with traditional fishing as suggested by Silas (1977) the traditional fishermen and their family members with a little training on seed collection and management of farm stock would greatly help to enhance oyster production and their earnings. 22.6% of operational cost of oysters can be reduced if their family members themselves can look after farm maintenance and predator eradication. As an experimental measure, fifteen fishermen families adopted oyster culture work under the transfer of technology programme on oyster culture and effectively managed 45 racks, which avocation yielded promising results and increased their annual income (Nayar *et al.*, 1979).

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PESTS AND PREDATORS OF OYSTERS

P. MUTHIAH[1], D. SUNDARARAJAN[1], G. SRINIVASAN[2] AND N. VAITHINATHAN[1]

INTRODUCTION

In all aquaculture practices the detrimental effects of cohabiting organisms are either by predation, competition, disease or parasitism. Hanson (1974) stated that limited predation can serve to weed out some diseased members of a crop and also help in controlling epizootic infections. But large-scale mortalities result in economic loss by reduction in the tended stock. Control of predation also means additional expense on the production cost (Mackenzie, 1970a). While evolving culture methods for fish or shellfish, identifying and proper use of methods to prevent and control numerous predators of cultivable organisms is absolutely essential to maximise production. In spite of having evolved control measures in oyster culture, developed countries like N. America, Britain and France still face periodical predator problems of serious threat to the stock under culture. In India, while conducting a series of experiments in the culture of oysters by rack and tray method at Tuticorin during 1977—1984 we have come across some problems of predation and competition in the oyster farm. Very often several transplanted oysters of size ranging from 25-85 mm were found dead in the growing trays in which were found large numbers of live gastropods (Pl. I a) later identified as *Cymatium cingulatum* (Lamarck). This led us to suspect the possibility that these might have been responsible for the mortality of oysters. Subsequent observations confirmed this.

This chapter chiefly describes the predatory role played by *Cymatium* and the extent of damage done to the tended stock.

BRIEF REVIEW OF COMMON PESTS AND PREDATORS

Among algae, *Gracilaria* sp. grows densely on the oyster cages kept in the farm which indirectly affects the regular water flow inside the cages. Boring sponges and clams are very rare and mortality of oysters due to these are not seen at Tuticorin. Occurrence of polyclad turbellarians on the spat settled on cultches and inside dead oyster spat are also noticed in the farm. But the intensity of its predation in Tuticorin Bay is negligible. Lunz (1947) found that oysters heavily infested with *Polydora* sp. were often poor in quality. Medcof (1946) confirmed that such infection does not noticeably decrease the condition of oyster but affects the market value in half-shell trade because of mud-blisters giving disagreeable appearance. *Polydora ciliata* and *P. armata* have been noticed in Athankarai estuary. Such a possibility at Tuticorin was effectively overcome by resorting to off-bottom culture and also periodical cleaning of oyster shell surface. Since it was feared that *Balanus* spp. settled on oysters and teak poles of the racks would compete with oysters for food and space, periodical cleaning helped to minimise the settlement. Slipper-shell *Crepidula fornicata*, notorious for causing mortality among young oysters, was totally absent in the farm area.

Lunz (1947) identified crabs as one of the most probable serious predators of oysters in 5 to 30 mm size. In the oyster farm at Tuticorin though some of the spat settled on tiles and rens were destroyed by crabs *Scylla serrata* and *Pagurus* sp. the loss due to this was negligible. The presence of commensal crabs among the oysters grown at Tuticorin was negligible.

Predation by starfish also was not noticed at Tuticorin Bay, apparently there is no population of sea stars in the surroundings. Mackenzie (1970 b) stated that if starfishes are present more than 1/m² the oyster stock would be reduced to non-commercial level. Predation by fishes and birds did not arise in the bay area perhaps due to the non-occurrence of these predators in considerable numbers in the bay.

Predatory gastropods known as oyster drills are considered as the deadliest among the enemies of oysters. In this country *Thais radolphii* has been

Present address : [1] CMFRI, Research Centre, Tuticorin-628 001.
[2] CMFRI, Research Centre, Madras-600 105.

noticed to bore young oysters at Athankarai estuarine region (CMFRI, 1974). In the oyster farm at Tuticorin, *Cymatium cingulatum* caused 13% of mortality of farm oysters (Thangavelu and Muthiah, 1983). Therefore pointed attention was paid to tackle this problem.

Mode of Attack

The gastropod gains entry into the trays and by remaining near the oysters or sometimes on the oyster shell, it introduces the proboscis when the valves are slightly open. The pleurembolic proboscis, having permanent sheath, functions due to muscles of the wall best suited to feeding on 'material not immediately accessible'. The jaws consist of two thin chitinous subtriangular plates (Pl. I b) having numerous longitudinal rows of scales. The jaws are lateral and appear to aid in opening the proboscis during feeding. An anaesthetizing fluid is injected on to the tissues of the unwary prey. Houbrick and Fretter (1969) found that the fluid which is exuded from the mouth of *C. nicobaricum* is acidic (pH 2.0). Day (1969) reported that the pH of pure secretion from proboscis gland of cymatiid *Argobuccinum argus* is 1.1. The acidic secretions poured into the oyster may create optimal conditions for certain enzymatic activities. In this case also a toxin similar to neurotoxin tetramine found in cymatiid *Fusitriton* (Russell, 1965) might have been employed by *C. cingulatum* in narcotising the oysters. Later the flesh is torn due to action of radula. Day (1969) pointed out that presence of calcium carbonate dissolving mechanism in cymatiids indicated that they are also able to drill the shell for feeding on bivalves.

Relation Between Size of *Cymatium* and the Radula

The radula is of taenioglossan type (Pl. I c). In *C. cingulatum* of size ranging from 21 mm to 72.5 mm in shell length, the radula varies from 2.07 to 5.35 mm in length and 247 μ to 600 μ in width. The study disclosed that within the size range of 21 to 73 mm increase in shell length was accompanied by a proportionate increase in radular dimensions. There is a close correlation between the length of radula and the shell length ($r=1$) eventhough radular dimensions vary sometimes among the individuals of the same shell length.

Size Relationship Between *Cymatium* and Oysters Preyed

Cymatium cingulatum attacked oysters of size 25 to 85 mm and the modal size of oysters killed was 53.3 mm (Thangavelu and Muthiah, 1983). Nearly 75% of mortality occurred in the size group of 40-65 mm. In order to find out the relationship between size of oysters preyed and size of *Cymatium*, 100 oysters of size 35 to 88 mm with *C. cingulatum* of known size were put in 12 box-type cages. Each cage was observed at fifteen days interval. From Table 1, it could be

Table 1. *Relationship of size of Cymatium and mean size of oysters preyed*

Size of *Cymatium* (mm)	Range of oysters Preyed (mm)	Mean size of oysters (mm)
45.0	39.0—64.0	49.7
52.0	34.0—64.0	48.6
52.0	39.0—65.0	56.2
52.5	49.0—84.0	59.4
58.3	53.0—86.0	68.0
58.5	49.0—84.0	69.4
63.2	41.0—84.0	54.4
64.0	39.0—70.0	55.0
65.6	42.0—83.0	63.8
69.0	37.0—68.0	54.0
72.5	35.0—80.0	64.7
74.0	47.0—88.0	68.5

seen that 45 mm *C. cingulatum* preyed upon oysters of 39 to 64 mm with a mean size of 49.7 mm. The mean size of oyster increased to 68.5 mm for the *Cymatium* of 74 mm in shell length. The correlation coefficient ($r=0.5$) shows fair degree of relationship between the size of gastropod and the size of oysters preyed upon.

Relation Between Oyster Stock and Predator Population

During 1978, totally 320 *C. cingulatum* were removed when the oyster stock in the farm was about 3,00,000. During 1980, when the stock increased to 6,00,000 the number of this predatory triton in the farm area swelled to 4,500 (Table 2). As Hanson (1974) pointed

Table 2. *Number and Size of C. cingulatum collected from Oyster farm in* 1980

Month	Number	Size (mm)		Mean (mm)	Mode (mm)
		Min.	Max.		
July	20	9.0	39.6	23.5	21.6
August	1497	7.5	71.2	34.22	38.45
September	2315	6.5	80.5	30.2	27.0
October	238	19.5	65.5	42.69	42.83
November	146	16.4	68.0	40.0	42.9
December	29	18.2	65.0	47.5	45.0

out the predators are appearing to be increasingly attracted with the prey abundance, thus aggravating the problem of predation.

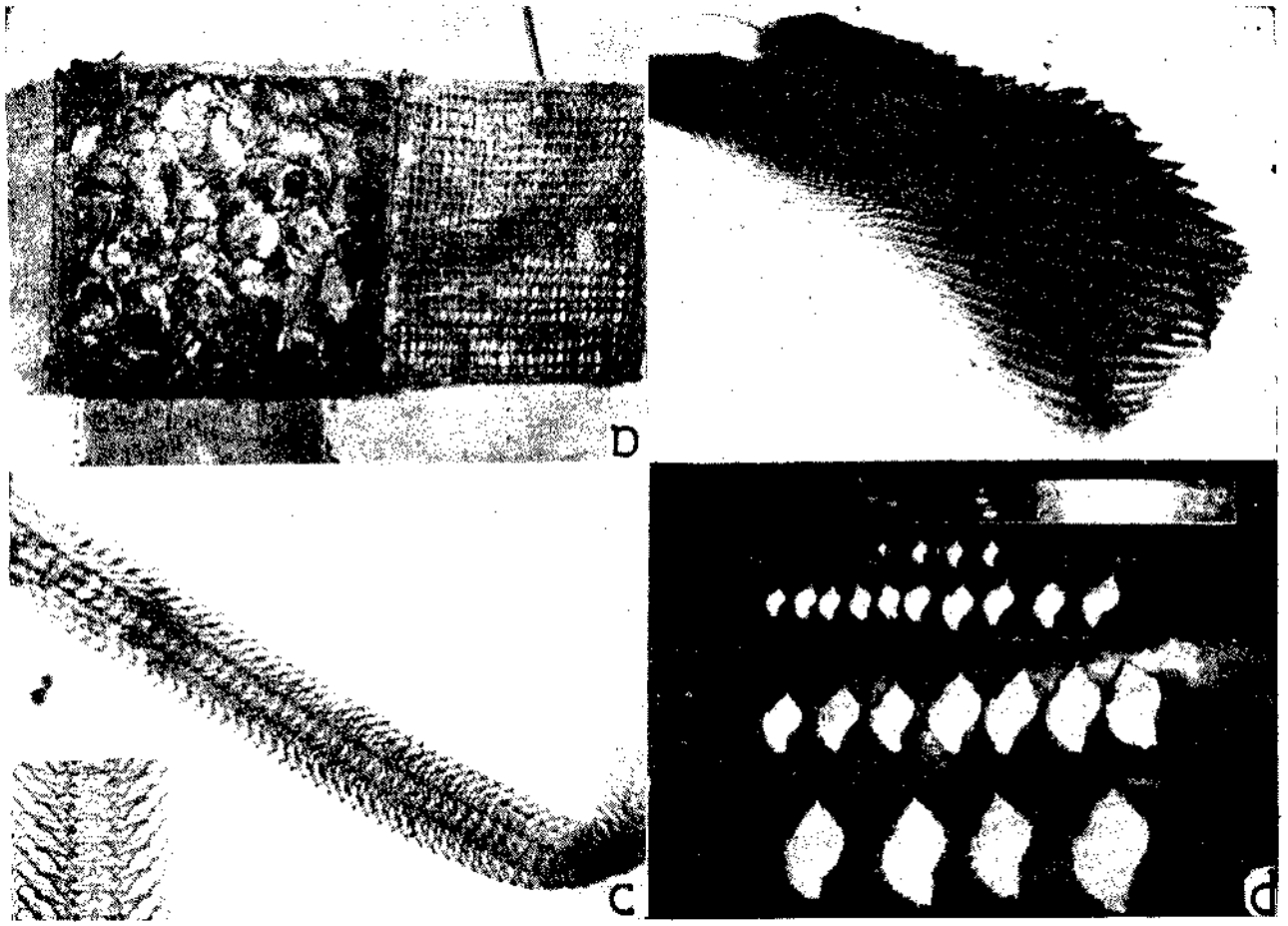

PLATE I. *a Cymatium cingulatum* inside oyster rearing cage; *b* Jaw plate; *c* Radula with portion enlarged; *d* Different sizes of *C. cingulatum*.

Growth Rate

The period of occurrence starts from late July to the end of December. The number increased from 20 in July, 1980 to 2,315 in September and decreased to 29 in December, 1980. Size ranges, mean and mode for *C. cingulatum* collected from the oyster cages during the months of July to December, 1980 are presented in Table 2. From this, it may be observed that the mode for July, 1980 was 21.6 mm which increased to 38.45 mm in August. In September it decreased to 27 mm and in the subsequent month it reached 45 mm. The size distribution curve (Fig. 1) was drawn from the measurements taken on the individuals occurring at the same site. The irregular progression of mode indicates that at most more than one year group is present. Because of their cannibalistic behaviour, the individual growth rate is difficult to follow. The wide range of size (Pl. I d) possibly suggests an extended breeding season as Thomson (1973) noticed in *Morula marginalba*. The maximum size of *C. cingulatum* collected from the farm was 80.5 mm, higher than *C. nicobaricum* (7.6 cm) recorded by Howbrick and Fretter (1969). The growth of *C. cingulatum* seems to be more rapid than *Urosalpinx cinerea* observed by Cole (1942) and growth of *M. marginalba* reported by Thomson (1973).

General Remarks

Drills can be controlled by treatment with chlorinated benzenes which are toxic to drills (Loosanoff *et al.* 1960 a, b). Mackenzie (1970 a) standardized the polystream (a mixture of polychlorinated benzenes) treatment at the rate of 9.5 kl/ha. Since the chemical treatment does not appear practical in the farm, elimination by handpicking was resorted to. During the season of its occurrence, all cages were constantly examined by employment of labour task force and the predators were removed. While future research should develop methods for economical and non-labour intensive treatment emphasis should also be paid on ways to prevent predation.

An important factor in the spread of diseases and predators in cultured shellfish population is through transfer of seed stock to growing areas. Japanese oyster drills *Ocenebra japonica* and carnivorous flat worms were imported to United States (Galtsoff 1964, Hanson 1974). Korringa (1942) described how *Crepidula* sp. was brought to Europe. Werner (1948), Chapman and Banner (1949) described the introduction of *Crepidula* sp. along with import and relaying of oysters in Germany and America respectively. These studies point out that very often spread of predators is unwarily done due to import of seed stock from one geographic area to another.

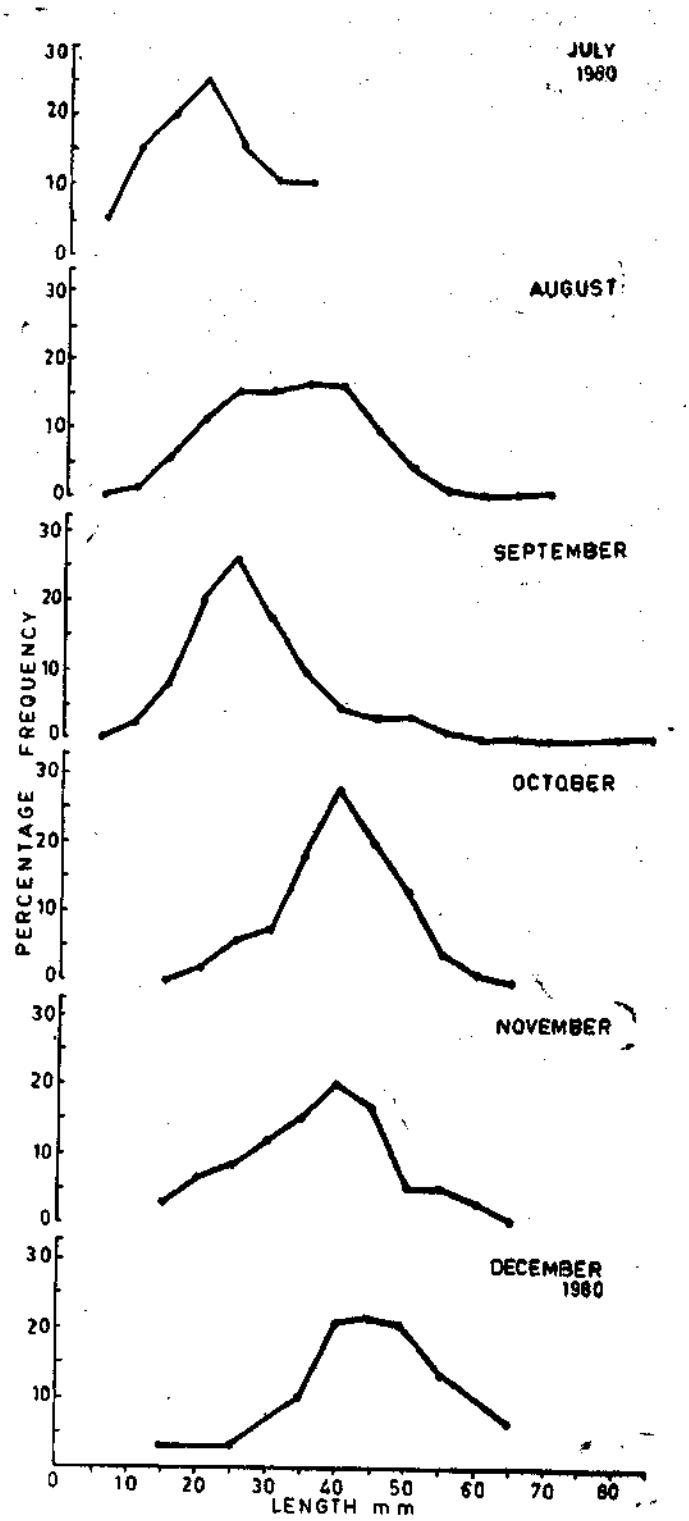

Fig. 1. Size distribution of *Cymatium cingulatum*

Controlling of predators means additional cost in the production of oysters. Mackenzie (1970a) gave that average cost of treating one acre of bottom with polystream for eradication of predatory gastropods was about $ 40 a year. In culture operations which involves crowding the animals, density problems of this nature are bound to arise and it becomes unavoidable to earmark a portion of the capital investment in extensive culture systems to achieve maximum production.

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13

OVERVIEW OF OYSTER CULTURE : PRESENT STATUS AND PROSPECTS

P. S. B. R. JAMES

The world oyster production is about one million tonnes per annum and the leading oyster poducing countries are U.S.A., Japan, Republic of Korea and France. There is world wide interest in increased supplies of oysters as the shellfish is a delicacy and command a high price. As in the case of other bivalve molluscs culture practices have to be adopted for augmenting production of oysters from natural beds for effecting regular supplies without depleting the natural resources. In view of this, oysters are cultured in several western countries and some countries in the Far East. As much as 12% of world production from aquaculture is accounted for by oyster culture.

OYSTER CULTURE IN THE WORLD

It is interesting to find that as early as First century B.C. the Romans were aware of the habits of oysters and employed simple methods for setting of oyster seed on piles fixed in coastal waters and reared them to a size when they were collected and consumed.

Japan has a long history of oyster culture carried out as an occupation since the seventeenth century to meet the large demand in that country (Cahn, 1950). Until the early part of this century simple culture methods were in vogue. Oyster spat were made to set on stones or sticks and they were reared on the spat collectors themselves or scattered at the bottom of shallow coastal areas. In 1923, Japanese scientists evolved the technique of hanging method of culture. This method gave consistently good production and was progressively adopted by commercial oyster farmers in several Prefectures including Kanagawa, Hiroshima, Miyagi, Iwate, Shizuoka, Wakayama, Shimane, Kumamoto and Mie (Imai, 1977).

Japan produces about 200,000 tonnes of oysters annually and the bulk of the oyster production is obtained in raft culture carried out in coastal waters in the depths of 3-9 m (Ikenoue, 1983). Rack culture is practised in shallower depths of 2-4 m. In long line culture, paired horizontal ropes are floated at the surface of the sea with the help of floats and anchored firmly at the ends. This method is successfully used in the open sea up to a depth of 30 m on the northern Pacific coast of Japan. The production from long line method ranks next to that from raft culture. From the rafts, racks or long lines ropes with spat collectors are hung. The culture techniques are being improved from time to time by using more durable materials like synthetic or steel ropes instead of straw ropes, and styrofoam floats in the place of wooden barrels used in earlier decades. The traditional bottom culture is rarely practised in Japan. Apart from oyster production for the local market, Japan produces and exports large quantities of seed of the Pacific oyster, ***Crassostrea gigas*** to U.S.A. and France for culture purposes.

U.S.A. ranks first in oyster production and the annual yield is about 300,000 tonnes of which 40% is from culture and the rest from harvesting from natural beds on the east and west coasts. Owing to the high cost of labour and materials involved in other types of culture, bottom sowing method only is used for culture of oysters in U.S.A.

Oyster production of the Republic of Korea was very low in 1950s but as a result of large scale raft culture, the production increased to 72,731 tonnes in

1972 and 187,033 tonnes in 1980. Korea exports canned oysters to U.S.A. and other countries.

In France, after several years of intensive studies, the technique of collection of oyster spat on lime-coated curved earthernware tiles was developed in the early period of this century. This type of spat collector is still in use in France and other countries like U.K. The annual oyster production of France amounts to 112,000 tonnes. Oyster culture is carried out by table method or bottom culture. The European oyster, *Ostrea edulis* is farmed by both bottom culture and table method while *Crassostrea gigas* is cultured only by the table method, rearing oyster seed in synthetic net bags called *pockets* laid over 'tables' erected in coastal waters. *C. gigas* is reared in plastic trays kept off bottom on steel framework int he Federal Republic of Germany (Meixner, 1979).

In Canada, different culture methods *viz.*, raft, stick as well as bottom culture are used for *C. gigas*. Apart from shell rens, sticks and veneer rings have been found useful for collection of oyster spat in British Columbia. Stick and tray culture methods are employed in Australia for culturing the Australian oyster, *C. commercialis* (Glude, 1979).

Oyster Culture in India

Small scale bottom culture of oysters by transplanting the spat from the natural beds to shallow areas of convenience has been in vogue in some places along the west cost like Jaytapur and Utsali but the production is insignificant. Hornell (1916) conducted experiments on spatfall of *Ostrea* (=*Crassostrea*) *madrasensis* on lime-coated tiles in Pulicat Lake. He also indicated the possibilities for the culture of oyster along the east coast using methods similar to those developed in Arcachon, Southern France (Hornell, 1910). Subsequently, some attempts for the collection of spat of *C. madrasensis* were made in the Madras Fisheries Department.

After experimenting with different methods of oyster culture, the rack-and-tray method for rearing of *Crassostrea madrasensis* was developed by the Central Marine Fisheries Research Institute at Tuticorin. The technique can be used in shallow coastal waters with water depth of about 2 m. Oyster spat are collected by laying lime-coated tiles or oyster shells in the vicinity of breeding oysters and they are reared in steel framed and synthetic twine meshed trays which are kept on a rack constructed with wooden poles (Mahadevan *et al.* 1980, Nayar and Mahadevan 1983). The oysters grow fast and attain an average size of 80-90 mm weighing 100-120 g with meat forming 8-10% at the end of one year. From culture operations in a 3-year period in 0.25 ha area, the estimated production of oyster would be 125 tonnes with a meat yield of 10 tonnes. At the end of each year approximately 42 tonnes of oyster could be harvested. Apart from meat, the oyster shells fetch a substantial return as by-product since they are used in the manufacture of Calcium carbide and cement. Spatfall and good growth of *C. madrasensis* has been observed in the estuarine environment at Athankarai (Rao *et al.*, 1983). There is influx of freshwater in the estuary in the northeast monsoon which causes mortality of oysters and therefore they have to be harvested before October. Experimental culture of *C. madrasensis* has been carried out in Mulki estuary and Cochin backwaters. Rapid growth of oysters has been recorded in Mulki estuary which is encouraging. The biological aspects such as age and growth, spawning, sex change, early development, spatfall, factors influencing them, pests and predators have been studied in *Crassostrea madrasensis* and *C. gryphoides*. Biological information on oysters in other areas is required for determining the scope for culture.

A new dimension has been given to oyster culture by the successful production of seed of the oyster *Crassostrea madrasensis* on mass scale by hatchery techniques developed at the Tuticorin Research Centre of CMFR Institute (Nayar *et al.*, 1984). The achievement has opened up avenues for supplying oyster seed at any time of the year for stocking in oyster farms.

There appears to be a gap in the proper assessment of the oyster resource potential of maritime States other than Tamil Nadu and Karnataka. It is necessary to take up the task by undertaking time bound programme of resources survey in the States and Union Territories.

Another line of future investigation pertains to the identification of suitable areas and sites for undertaking extensive culture of edible oysters. Some data base for site selection is available only for Tamil Nadu. The advances made in oyster culture technology have evoked widespread interest amongst several entrepreneurs and agencies in States like Kerala, Goa and Maharashtra. Unfortunately, precise information on the suitability of sites in these states is lacking. This lacuna in our knowledge should be filled.

It is not uncommon to come across instances of large scale oyster mortalities both epizootic and enzootic in culture as has been the experience in coun-

tries like U.S.A., France and the Netherlands. But to a certain extent such problems have been overcome by diagnosing the aetiological agents and taking up preventive measures and treatment. In some countries like U.S.A., disease resistant strains have been developed by spotting oyster disease affected areas and locating the surviving oysters in the midst of large scale destruction. These oysters are bred in captivity and the progeny are broadcast over natural beds so that the stock will be disease free due to acquired immunity. Cross breeding between an oyster strain known to be disease resistant with another of established susceptibility to disease would help to evolve one which is totally disease resistant. Large scale mortality of oysters on account of diseases of viral and other microbial origin are not common in India. Identification of superior strains of oysterwith reference to faster growth, disease resistance, higher yield of meat involving genetic selection and cross breeding would be a future line of work in this country.

The technology of oyster farming followed in advanced countries is capital intensive. Our efforts, on the other hand, have been directed towards development of low cost technology. The oyster meat at present commands a lucrative price at international level. Experimental culture undertaken at Tuticorin showed that the present rack and tray culture system can be further modified and made cheaper by reducing the cost on labour and material inputs if string (ren) culture is taken up. This would perhaps also serve as a feasible alternate method for areas along our coasts which do not have shallow tidal flats. Development of culture technology suitable for the geographical and topographical characteristics of each maritime zone will ensure greater success.

The labour intensive spat collection and transportation method by using lime coated tiles has been found to be costlier than collecting the spat on shell cultch. Large scale spat collection by using shells would not only reduce the expenditure but enable long line culture in deeper areas.

The future development of increased oyster culture activities also depends on finding potential new markets. Initial test marketing indicated good demand within the country. The export market for oysters is already known but the quality of the product in competition with other exporting countries would be a deciding factor.

Paralytic shellfish poisoning(PSP) represents a serious health hazard which needs attention of oyster culturists since the oysters are filter feeders ingesting toxic dinoflagellates like *Gonyaulax* (Ray, 1984). Further, it is well known that oysters tend to accumulate heavy metals like mercury and zinc beyond the acceptable level of safety. It would, therefore, be necessary to strictly enforce pollution control measures in the areas of oyster culture.

Research and Development

Future research priorities include investigations on nutrition, growth, improvement of stocks by selective breeding, utilization of other species of oysters that are suitable forcultivation and development of techniques for monitoring and control of marine toxins.

For further development of oyster culture on a commercial scale, coastal tracts have to be made available to prospective farmers on lease. For this purpose, the Government have to enact suitable legislation. However, the allotment of coastal areas for oyster farming should not at the same time come into conflict with traditional fishing activities.

Several essential inputs are needed for oyster farming. Financial institutions like banks and agricultural and cooperative credit organizations have to extend financial assistance to those interested in the new avocation.

It is hoped that with the technical knowhow now available, entrepreneurs will come forward and take up oyster farming so that the resources available along our coasts are fruitfully utilized to augment marine fish production. The Institute will extend technical expertise to such ventures. The feedback data obtained from commercial ventures would help the scientists to improve and streamline the culture practices wherever necessary.

Extension activities have a special role in popularising oyster farming as well as making the oyster a relished sea food amongst the public. Training of intending farmers and entrepreneurs in oyster culture, popularization of oyster as food, educating the industry on post-harvest and processing technology and promoting internal and external markets would go a long way in establishing commercial oyster farming in the country.

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