The Marine Polyzoa of the Isle of Man

by

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"The zoologist finds his pleasure in the contemplation of their novel forms, in the examination of those characters which distinguish them as species, in the quest of their mutual affinities, their relations and analogies with other beings, the order in which Creative Wisdom may seem to have called them into existence, their habits, economy and uses"

Johnston (1838)
"A History of the British Zoophytes"

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INTRODUCTION

Nitsche (1870) proposed that the Polyzoa (= Bryozoa) should be divided into the Ectoprocta and the Entoprocta. The Entoprocta would contain the genera *Pedicellina* M.Sars, *Urnatella* Leidy and *Loxosoma* Keferstein, the rest of the Polyzoa would be included in the Ectoprocta. In the Ectoprocta the anus is situated outside the tentacular circlet, in the Entoprocta it is inside the lophophore. Hatschek (1888) raised the Entoprocta to phylum level but this view was not generally accepted and even today some authorities treat the Entoprocta as a sub-group of the Polyzoa. In 1921 A. H. Clarke recognized the non-coelomate nature of the Entoprocta and proposed placing them in a new phylum the Calyssozoa. Cori (1929) agreed that they were a separate phylum and suggested the term Kamptozoa. Both Calyssozoa and Kamptozoa are used by some contemporary authors but in this account the original term Entoprocta will be used and the group will be treated as a phylum distinct from the phylum Ectoprocta.

PART I

Phylum Entoprocta

Introduction.

The Entoprocta are divided into three families, the fresh-water Urnatellidae, and the Loxosomatidae and Pedicellinidae. Members of the Loxosomatidae and the Pedicellinidae are present in Manx waters. Before the present study only <u>Loxosomella fauveli</u> Bobin and Prenant, <u>Pedicellina cernua</u> (Pallas) and <u>Barentsia gracilis</u> (M.Sars) had been recorded from the Isle of Man.

Material and Methods used in the present investigation.

Between October 1959 and June 1962 large amounts of supports suitable for entoproct colonization were examined. This material included large numbers of polychaete worms (including 28 <u>Aphrodite aculeata</u> L., 18 <u>Hermione hystrix</u> (Savigny) and many other Polychaetae Aphroditidae), many and various worm tubes (particularly those of <u>Chaetopterus variopedatus</u> (Renier)), 41 <u>Phascolion strombi</u> (Montagu), large numbers of various entoprocts and hydroids, some ophiuroids, crinoids, sponges and ascidians and large quantities of dead shell. Most of the material was dredged off the south of the Isle of Man, but some, including specimens of <u>Aphrodite aculeata</u>, was trawled E. of Langness (by A. B. Bowers) and in Luce Bay (by D. J. Symonds). Suitable materials from the "Faunal Collections" at Port Erin were also examined, this source provided 23 of the <u>Phascolion strombi</u> examined. All fresh material was examined under a binocular microscope and observations made on the living Entoprocts. The material was then stored in alcohol or, better, formalin. No specimens were stained, nor were any sections cut. Identifications were made under the high magnifications of a monocular microscope. Specimens were measured by using an eye-piece micrometer. In all 16 species were collected and are listed below with notes on their occurrence:

Family Loxosomatidae.

Genus <u>Loxosoma</u> Keferstein.

<u>Loxosoma</u> sp.nov.

Genus Loxosomella Mortensen

Loxosomella obesa Atkins	New record for Irish sea.
L.murmanica (Nilus)	New record for Irish sea.
L.nitschei (Vigelius)	New record for Irish sea.
L.compressus Nielsen & Ryland	New British record.
L.fauveli Bobin & Prenant	Previously recorded from Isle of Man.
L.arvyae Bobin & Prenant	New British record.
L.bouxini Bobin & Prenant	New British record.
L.fungiformis Bobin & Prenant	New British record.
Loxosomella sp.nov.	
L.marsyops Nielsen & Ryland	New British record.
L.claviformis Hincks	Widely distributed in N. W. Europe.

Family Pedicellinidae

Genus Pedicellina

Pedicellina cernua (Pallas)	Widely distributed.
P.nutans Dalyell	Widely distributed.

Genus Barentsia

Barentsia gracilis (M.Sars)	Widely distributed.
Barentsia sp.nov?	

In addition to these species it is quite likely that further entoproct species would be found in Manx waters if the correct supports were searched. The data obtained from each of the above species are discussed below.

FAMILY LOXOSOMATIDAE

Diagnosis

Solitary marine, often epizoic Entoprocts in which the calyx is never separated from the peduncle by a diaphragm and is never deciduous. Peduncle rarely absent, usually more or less long and often terminating in an adhesive attachment. An adhesive-producing gland may be present in the peduncle at some stage in the life-history. Buds develop on the anterior face of the calyx but soon separate from the parent and do not form colonies but some species by rapid fixation of the buds soon after separation, form dense populations.

Of the four genera (Loxosoma, Loxosomella, Loxocalyx and Loxomespilon) included in the family species of only Loxosoma and Loxosomella have been collected in Manx waters.

GENUS LOXOSOMA Keferstein 1862 (emend. Mortensen 1911)

Diagnosis

Foot developed as sucking disk, peduncle with straight and oblique muscles. No foot gland, only single gland-cells on the disk. Animal able to change position.

The only Loxosoma collected appears not to have been previously described.

Loxosoma sp.nov.

Collection

About 30 specimens were found on the inside of an unidentified, arenaceous worm-tube dredged 5 mi. N 46° W of the Chicken Rock, depth 28 fm. (50.5 m) on 12.4.61.

Description (See Fig I.1)

This description is taken from 6 specimens in semi- to wholly contracted states after preservation in 5% formalin for some weeks.

This a large species measuring up to 1.2 mm. total length of which the calyx measures about half. The calyx, clearly demarcated from the peduncle, has the shape of an inverted pear. 16 - 21 long tentacles have been counted. Sensory papillae are absent but what are probably lateral sence organs can be seen under high power magnification. These consist of a small pit with a few short cilia and are well supplied with nerve and muscle fibres. There are no trace of 'flask organs' such as are found on L.davenporti Nickerson, L.loxalina Assheton and L.saltans Assheton. The tri-lobed stomach is slightly wider than long. The thick-walled intestine leads into the rectum which terminates at a conspicuous anal papilla reaching to about the middle of the lophophore. The budding zone is anterior and slightly proximal to the top of the stomach. No large buds were

Table 1. Dimensions of 6 specimens of Loxosoma sp.nov. Units are μ.

Dimension	Specimen 1	2	3	4	5	6	Mean
Total length	1188	1012	1012	924	915	853	984
Peduncle length	616	484	528	396	396	378	466
Maximum width	405	369	350	326	360	405	371
Peduncle width	141	132	132	123	149	132	134
Stomach length	141	123	132	141	132	132	133
Stomach width	149	132	149	158	149	158	149
Tentacle number	20	20	18	16	21	20	-
No. of buds	1	-	2	1	1	2	-

Table 2. Proportions of Loxosoma sp. nov. based on measurements of six specimens.

Ratio	Mean	Range
Calyx Length: Peduncle Length	1.10	0.91-1.33
Calyx Length: Calyx Width	2.75	2.54-2.93
Calyx Width: Peduncle Width	2.70	2.41-2.87
Stomach Length: Stomach Width	.87	.76-.95

present. Most specimens bear one or two tiny buds, usually one on each side. The calyx is narrow in profile and the parenchyma is not dense. The peduncle has well-defined longitudinal muscles some of which become oblique and cross the peduncle level with the stomach. There is a slight development of oblique muscle in the foot region. A few longitudinal and circular fibres can be seen in the calyx. The pedal disk is well developed. A row of large, square cells (similar to those described in Loxosoma davenporti Nickerson, L.loricatum Harmer, Loxosomella crassicandata (Salensky), and Loxocalyx tethyae (Salensky) runs along the dorsal surface of the peduncle to the level of the stomach where it merges with and becomes indistinguishable from the other cells of the dorsal surface.

The dimensions of the 6 specimens (semi-contracted and in formalin) measured are summarised in Table 1 and 2.

Live specimens are very active and when disturbed sway in all directions. The calyx can be rotated through 180° relative to the foot. No specimens were observed to detach themselves from their support but when detached with a needle specimens quickly and easily reattached themselves.

Discussion

The absence of a pedal gland in the adult (no large buds were seen and therefore the condition in the bud is unknown) and the ability of the specimens to reattach themselves to supports place this species in the genus Loxosoma. In this genus it has obvious affinities with the group of species L.pectinaricola Franzen, L.davenporti, L.loxalina and L.saltans. It differs from the last three in the absence of flask organs but does not say what proportion are without them; it seems unlikely that all of 30 specimens collected would be without these organs. Other similarities with L.davenporti are in the row of large cells along the dorsal surface of the peduncle (which also occur in L.saltans, L.loxalina, etc.), the position of the anal papilla and the position and shape of the stomach. It differs from L.davenporti in the absence of flask organs, the absence of lateral alae in the contracted state, and the tentacle number (18 - 29 in L.davenporti) and in the absence of large numbers of buds (this may be a seasonal effect). L.pectinaricola has fewer tentacles (14 - 16) than the present species and lacks the row of large cells on the peduncle.

GENUS LOXOSOMELLA Mortensen 1911

Diagnosis

Foot gland present only in the bud, atrophying after secreting a fluid which cements the animal to the support for life. The adult peduncle may be more or less expanded into a pedal disk.

A list of the Loxosomella species recorded from Manx waters is given on p.2 . For other N. W. European species see Bobin and Prenant (1956), Ryland (1960, 1961), Ryland and Nielsen (1961) and Franzen (1963).

Key to the Manx species of Loxosomella

1. Large species, up to 2mm. lophophore small in relation to calyx. Calyx often greatly swollen, budding zone immediately below lophophore

L. obesa

Usually smaller than 1.5 mm., calyx not enlarged nor stomach greatly swollen. Lophophore normal size relative to calyx 2

2. Compressed laterally, single median budding zone. Among dorsal spines of polynoid worm

L. compressa

Not compressed laterally, two lateral budding zones 3

3. Inside worm tubes 4

Not inside worm tubes, on ectoprocts, polychaetes or sipunculids 5

4. Highly contractile peduncle attached in pocket of worm tube lining. Calyx rounded

L. marsyops

Very short peduncle terminating in pedal disk, not in pocket of worm tube lining. Calyx with obvious lateral sense organs on "shoulders"

Loxosomella sp.nov.

5. 8 tentacles only (rarely more) 6

More than 8 tentacles 7

6. Frequently with secondary brown 'cuticle' covering part of peduncle. Stomach wider than high, relatively small and well separated from body wall. Relatively small lophophore, budding zone above stomach. On <u>Phascolion strombi</u>

<u>L. murmanica</u>

Rarely with secondary 'cuticle' lophophore normal size. Stomach very wide in relation to its height and almost reaches body wall, and with well marked lateral pouches. Budding zone level with stomach. On <u>Phascolion strombi</u> and ectoprocts

<u>L. nitschei</u>

7. On <u>Aphrodite aculeata</u> or <u>Hermione hystrix</u> 8

On <u>Phascolion strombi</u> 9

8. Peduncle and calyx not clearly demarcated, animal club-shaped. Large, well-defined pedal disk, 8-13 (usually 11) tentacles

<u>L. claviformis</u>

Not club-like. Peduncle and calyx clearly demarcated. 8-11 tentacles. Peduncle shorter than calyx and with a small pedal disk

<u>L. fauveli</u>

9. The large, ovoid, stomach, slightly higher than wide, may extend to body wall and is tangential to the lophophore. No lateral pouches but superior and lateral stomach walls are very thick. 8-12 (usually 10-11) tentacles. Buds level with base of stomach and may be directed laterally or towards base

<u>L. bouxini</u>

Stomach of median dimensions, about round, a little wider than high but well separated from body wall. 8-10 short tentacles.

<u>L. arvyae</u>

Stomach thick-walled, much wider than high, 9-10 tentacles. Fungiform.

<u>L. fungiformis</u>

<u>Loxosomella obesum</u> Atkins (Fig I 2)

<u>Description</u>

Large (maximum length recorded at Port Erin 1.8 mm., at Plymouth

Table 3 . Dimensions of <u>Loxosomella obesa</u>. 50 specimens measured. Units are μ.

Dimension	Mean	Range
Total length	979	546–1795
Peduncle length	398	211–748
Calyx length	601	335–1020
Maximum width	429	309–606
Peduncle width	129	70–158

Table 4. Proportions of <u>Loxosomella obesa</u> based on measurements of 50 specimens.

Ratio	Mean	Range
Calyx length:Peduncle length	1·55	1·10– 2·50
Calyx length:Calyx width	1·41	0·9 – 2·0
Calyx width:Peduncle width	3·6	1·2 – 8·9
Peduncle length:Peduncle width	3·1	1·1 – 5·0

2.4mm.). Small circular lophophore with 8 tentacles. (rarely 9 at Plymouth (Atkins 1932)). Calyx clearly separated from the peduncle and often greatly swollen by enlargement of parts of the stomach. In small specimens the calyx is fairly narrow below the lophophore. Calyx equals $\frac{1}{3}$ - $\frac{1}{2}$ total length in large specimens. Only longitudinal muscle fibres in stalk. Pedal disk slightly developed. Vestiges of pedal gland may be visible. Budding zone immediately below lophophore. Numerous buds - up to 6 each side - often present. Up to 26 embryos may be present in vestibule (Atkins 1932).

As noted by Atkins there are 3 types of individual. 1. Without swollen stomach, 2. Stomach swollen, calyx oval, 3. Stomach swollen, calyx globular. At Plymouth the peduncle of type 2 zooids equals $\frac{1}{2}$ - $\frac{2}{3}$ of their total length and that of type 3 is less than $\frac{1}{2}$ the total length. In Manx material the peduncle of type 3 is slightly shorter in proportion to total length than in type 2, but the difference is slight and in both type 2 and 3 the mean peduncle length is less than $\frac{1}{2}$ the total length.

Dimensions from 50 individuals are given in Table 3 ~~14~~ which includes very few type 1 individuals and about equal numbers of types 2 and 3

Discussion

Previously recorded only from Plymouth, *L.obesum* occurred on 7 (32%) of 22 *Aphrodite aculeata* from the Isle of Man and on 2 of 6 *A.aculeata* from Luce Bay (S. W. Scotland). At Plymouth it was present on 12.3% of 146 *A.aculeata* examined (Atkins 1932). It occurs only on the ventral surface of the posterior elytrae and on the posterior half of the dorsal surface of *A.aculeata*. Over 100 *L.obesum* have been noted on a single *A.aculeata*.

Buds were observed in April, May and June the only months in which *L.obesum* was collected.

Loxosomella compressa Nielsen and Ryland (Fig 13)

Description

The mean length of Manx material is ca. 500μ., maximum size

Table 5 Dimensions of Manx and Norwegian (from Nielsen and Ryland) specimens of Loxosomella compressa. 16 Manx specimens and 20 Norwegian specimens measured. Units are μ.

Dimension	Manx Mean	Manx Range	Norwegian Mean	Norwegian S.D.
Calyx length	198	141-263	178	28
Peduncle length	298	184-528	267	78
Total length	496	352-704	445	91
Peduncle depth	84	52-114	78	12
Calyx depth.	150	132-211	120	14

Table 6 Proportions of Manx Loxosomella compressa basedon measurements of 16 specimens.

Ratio	Mean	Range
Peduncle length:Calyx length	1·5	1·0 - 3·0
Calyx length:Calyx depth	1·4	1·1 - 1·9
Calyx depth:Peduncle depth	1·9	1·3 - 3·6
Peduncle length: Peduncle depth	3·7	2·1 - 6·0

recorded 700μ. Calyx laterally compressed to about $\frac{2}{3}$ its depth 8 tentacles (rarely 9), lophophore slightly oblique. Sensory papillae absent. Stomach not tri-lobed. One or two buds occasionally present in median group about midway up calyx. Pedal gland present in large buds. Calyx passes abruptly into peduncle which is also slightly compressed. Peduncle, $1\frac{1}{2}$ - 2 times length of calyx, tapers towards base.

Table 5+6 includes the dimensions of 16 Manx specimens.

Discussion

The type material was collected by Nielsen and Ryland (1961) on the notopodial setae of the polynoid Lagisca extenuata (Grube) from the North Brattholmen Lophelia-reef near Bergen. About 20% of Manx L.extenuata searched bore Loxosomella compressa. Most of the worms were collected either in tufts of Cellaria fistulosa L. or Chaetopterus tubes dredged about 4 ml. (6.5 km) N. W. of the Chicken Rock (depth 27-30 f. = 49-55 m.) but some were dredged in 10 f. (18 m.) close to Port Erin Breakwater. Up to about 40 L.compressa may occur on a worm and all are attached to the notopodial setae.

Specimens were collected in September, October and November. In all the populations examined some specimens bore buds. A few individuals with embryos and well-developed gonads were present in October.

This species has previously been recorded only from the Norwegian type locality. I have collected it off both Manx and Northumbrian coasts.

Loxosomella marsypos Nielsen and Ryland (fig I4)

The original description of this species was based on only a few specimens. A large number of specimens have been collected from Manx waters. Manx specimens are larger than the type material; all dimensions in the description are based on Manx specimens.

Table 7. Dimension of *Loxosomella marsyons*. 50 specimens measured. Units are μ.

Dimension	Calyx	Peduncle			
		Relaxed	Slightly contracted	Moderately contracted	Fully contracted
	(50)	(13)	(16)	(10)	(11)
Mean length	253	363	277	276	222
Length range	167–396	238–510	176–387	238–370	176–290
Mean width	229	202	208	242	271
Width range	132–309	106–290	141–309	185–290	202–352

Table 8 Proportions of *Loxosomella marsyons*. Based on 50 specimens.

Ratio		State of Peduncle			
		Relaxed	Slightly contracted	Moderately contracted	Fully contracted
Calyx width : Peduncle width	Mean	1·12	1·06	·97	·92
	Range	·9–1·5	·8–1·4	·8–1·0	·7–1·0
Peduncle length : Peduncle width	Mean	1·96	1·3	1·14	·8
	Range	1·1–4·4	·8–1·8	·8–1·5	·7–1·2
Calyx length : Peduncle length	Mean	·75	·9	·94	1·2
	Range	·4–1·2	·6–1·6	·5–1·3	·9–1·6

Calyx length: Calyx width Mean 1·1 Range ·8–1·9

Description

Large fully relaxed individuals measure up to 800 μ in length. Highly contractile peduncle; when relaxed it equals $\frac{2}{3}$ total length, when contracted it is shorter than calyx. 8-12 long tentacles. Sensory papillae absent. Stomach deep, not clearly tri-lobed. Budding zone antero-lateral, near base of stomach. Pedal gland well-developed in large buds. Peduncle and foot characteristic of the species. The fully relaxed peduncle broadens abruptly just below calyx and tapers gradually to base, when slightly contracted a pair of subcircular lateral expansions are present below calyx, the fully contracted peduncle is basically coordate in form and cups the base of the calyx in its distal end. Peduncles in different states ofcontraction are illustrated in Fig I 4. No pedal gland in adult but one to three oval bodies occur in peduncle of some specimens, these may be vestiges of pedal gland of bud.

Dimensions of 50 Manx specimens are given in Tables 7 and 8.

Discussion

Previously recorded only from the type locality, the North Brattholmen Iophelia-reef near Bergen, where it occurs in the tubes of Eunice pennata (O.F.Muller) in 44-75 f. (85-130 m.) (Ryland Nielsen 1961). Off the Isle of Man L.compressa occurs in the tubes of Chaetopterus variopedatus. The majority of the 200 specimens collected were in Chaetopterus tubes dredged about $4\frac{1}{2}$ ml. (7.2 km.) N. W. of the Chicken Rock in 27-30 f. (50-55 m.). In both Manx and Norwegian material the entoproct occupies a characteristic site. The peduncle is attached to the lining of the tube at the apex of a small triangular pocket in the lining membranes, contraction of the peduncle withdraws the animal into the pocket. (See Fig I 5). Ryland and Nielsen found only small "colonies" of few individuals but Manx "colonies" are larger; up to 60 individuals have been collected from a single Chaetopterus tube. Fig I 5 gives a semi-diagrammatical indication of the density reached in some "colonies". Manx specimens measure up to 800 μ ., the largest specimen measured by Ryland and Nielsen was 370 μ long.

Table 9. Dimensions of Loxosomella sp.nov.. 20 specimens measured. Units are μ

Dimension	Mean	Range
Total length	316	194-440
Calyx length	242	150-328
Peduncle length	74	44-132
Calyx width	140	88-237
Peduncle width	70	44-79

Table 10 .Proportions of Loxosomella sp.nov.. Based on 20 specimens

Ratio	Mean	Range
Calyx length: Peduncle length	3·4	2·3-4·4
Calyx width: Peduncle width	3·0	2·1-3·6
Calyx length: Calyx width	1·3	1·0-1·7
Peduncle length: Peduncle width	1·1	1·0-1·5

The mature gonads are large and appear white by direct light. Under low-magnification they appear as small conspicuous paired, white spots. Specimens were collected in October and January; buds and well developed gonads were present in both months and embryos in October only.

Loxosomella sp.nov.

A description of this species will be published shortly and specimens will be deposited at the British Museum.

Description (Fig I 6)

Small. (Maximum length 440 μ). Peduncle much shorter than calyx. Large oblique lophophore, in life peduncle is bent so that lophophore inclines towards support. 10-14 tentacles. Calyx slightly longer than wide, and resembles inverted pear. Lophophore slightly narrower than rest of calyx. Lateral sense organs on conspicuous shoulder-like protruberances, on each side of and level with centre of lophophore. Short peduncle not clearly demarcated from calyx. Peduncle firmly attached by thin, often triangular, pedal area. Pedal gland absent in adult, well-developed in large buds. Budding zone anterior, level with top of stomach. Most specimens have one bud, some two. Heart-shaped stomach without lateral lobes. Up to 7 embryos occur in the atrial cavity. Ovaries lie between stomach and distal part of lophophore.

Dimensions and proportions of 20 individuals are given in Table 9 and 10

Habitat

Collected 4½ ml. (7.8 km.) at N 36° W of Chicken Rock. (Decca Co-ordinates Red E 13.65, Green E 45.95) in 27 f. (50 m.). About 30 individuals were found attached to the membranous inner lining of an arenaceous worm-tube.

Affinities

In general appearance this species is similar to L.fagei but it can be readily distinguished from this species by its greater tentacle numbers of 10-14 (8 in L.fagei), its heart-shaped stomach (almost round

Table 11 Dimensions of *Loxosomella murmanica*. 19 specimens measured.

Dimension	Mean	Range
Total length	331	228–440
Calyx length	210	141–317
Peduncle length	121	79–176
Calyx width	200	114–370
Peduncle width	61	44–88

Table 12 Proportions of *Loxosomella murmanica*. Based on 19 specimens.

Ratio	Mean	Range
Calyx length: Peduncle length	1·8	1·1–2·3
Calyx length: Calyx width	1·1	·8–1·7
Calyx width: Peduncle width	3·2	2·0–4·4
Peduncle length: Peduncle width	2·0	1·1–2·8

in L.fagei) and the different host (L.fagei has been recorded only on Hermione hystrix).

Loxosomella murmanica Nilus (Fig17)

Description

Small (Bobin and Prenant(1956) give maximum length 650 μ , Manx specimens reach 450 μ.) Peduncle usually shorter than calyx sometimes slightly longer. Peduncle muscular, thick, often arched in sagittal plane. Peduncle cuticle ridged, pedal disk slightly developed. Pedal gland in bud, absent in adult. Calyx, flattened antero-posteriorly, well-marked cuticular "epaulettes" often present near origin of calyx from peduncle. Calyx racquet-shaped. Lophophore is tangential to calyx and its outline may extend beyond the oval contour of the calyx. No visible sensory papillae. Small stomach, ellipsoidal and transversally elongated; lateral pouches absent. Distance between body wall and stomach usually equals about half stomach width. Budding zone between lophophore and stomach. Not more than 1 bud per sideusually present. Hermaphrodite. When well-developed eggs present calyx becomes distended and almost circular in outline including even the retracted lophophore within its outline.

The dimensions and proportions of 19 Manx specimens are given in Table 11 and 12

Discussion

L.murmanica occurs on Phascolion strombi. A brown "secondary cuticle", thought to be produced by the sipunculid host, often covers all or part of the peduncle and calyx of L.murmanica. This "cuticle" sometimes occurs on other entoproct species but is more extensively developed on L.murmanica and if present on a specimen indicates that it is probably L.murmanica. This "cuticle" sometimes partly or completely encloses the entoproct. Specimens in which the "cuticle" is extensive are often distorted.

L.murmanica is widely distributed. It has been recorded from Kola Fjord, Roscoff, Dinard, Concarneau, Arachon, Plymouth etc. In all cases it occurs on Phascolion strombi. In Manx collections it

Table 13 Dimensions of *Loxosomella nitschei* from Manx waters compared with those of specimens from Norwegian and Northumbrian waters. 40 Manx specimens measured. Data for 25 Norwegian and 20 Northumbrian specimens from Ryland (1961) Units are μ.

Material		Dimension				
		Total length	Calyx length	Peduncle length	Maximum width	Peduncle width
Manx	Mean	273	162	111	160	57
	Range	158–378	79–246	79–158	188–246	35–79
Norwegian	Mean	430	206	240		
	S.D.	42	22	39		
Northumbrian	Mean	490	240	284		
	S.D.	63	49	40		

Table 14 . Proportions of Manx *Loxosomella nitschei*. Based on 40 specimens.

Ratio	Mean	Range
Calyx length:Peduncle length	1·5	1·0–2·3
Calyx length:Calyx width	1·04	·7–1·5
Calyx width:Peduncle width	2·8	2·0–4·0

occurred on 14 (32%) of 41 P.strombi examined. (cf. Concarneau 57 (66%) of 87 P.strombi "infected" and Roscoff 100% "infection"). L.murmanica has been found mixed with populations of L.bouxini, L.nitschei and L.fungiformis. (See p 20 for discussion of entoprocts and P.strombi).

Buds were seen in April.

Loxosomella nitschei (Vigelius) (Fig. I 8)

Description

Calyx and contour less elongate than L.murmanica. Lophophore relatively larger and with stronger muscles than L.murmanica. When contracted lophophore lies within calyx outline but when expanded it modifies calyx contour to a regular oval. 8 (rarely 9) tentacles, when retracted have wider base than L.murmanica. Lateral sense-organs absent. Large, wide stomach almost as wide as calyx. Stomach length is about half its width but varies with state of contraction. The well marked lateral pouches are less conspicuous when the stomach is at its longest. Large pharynx obvious in anterior view. Budding zone level with top of stomach. In French material up to two buds occur on each side (Bobin and Prenant 1956), in Manx material only one bud has been seen on each side. Peduncle shorter than calyx. Cuticle thin and uniform but may be ridged on peduncle. Distance between stomach and body wall is always less than a quarter of stomach width.

Dimensions of Manx and Norwegian and Northumbrian material from Ryland 1961 are summarized in Table 13 and 14

Discussion

L.nitschei occurred on 28 (68%) of 41 Phascolion strombi examined. It occurs most often around the anus and on the posterior part of the sipunculid. It has been recorded on P.strombi from Dinard and Concarneau (Bobin and Prenant 1956). Manx specimens, like the French material, occur in mixed populations with other species e.g. L.bouxini, L.murmanica and L.fungiformis (see also p 20). The type material (Vigelius 1882), which has been lost, was described from the ectoproct Menipea ternata (Ellis and Sol.) from the Barents Sea. Roper (1913) and Harmer (1915) described material collected in Northumberland on ectoprocts, hydroids etc. Recently Ryland (1961) found this species on Dendrobeania murrayana (Johnston) and Callopora craticula (Alder) from the Raune Fjord near Bergen. Ryland's material is identical with Roper's but differs from French specimens (Bobin and Prenant 1956) in the pattern of budding. In N. Sea and Norwegian material the younger buds are below the older; French specimens have the younger buds above the older. Unfortunately none of the Manx specimens have more than one bud on each side. (I have recently (at the Dove Marine Laboratory Cullercoats) examined specimens (from hydroids, ectoprocts and shell fragments) indentical with those of Ryland, Roper and Harmer and am of the opinion that this is a species separate from that occuring on Phascolion strombi from Manx, Northumbrian and French coasts.)

Buds were present on Manx material collected in May and in specimens from the Port Erin Faunal Collections dated 1.8.46. Buds were present on Norwegian material in May.

Loxosomella claviformis (Hincks). (FigI 9)

This is a very variable species and different forms have been described severally as separate species or as varieties of L.claviformis. Manx specimens are only of one type which is described below. For a discussion of the variation of L.claviformis see Atkins (1932) and Bobin and Prenant (1953 and 1956).

Description

The Manx L.claviformis are of the type designated L.claviformis var. by Bobin and Prenant 1956.

Up to 700μ in total length. Club-like, no clear demarcation between calyx and peduncle. Most claviform when tentacles retracted. Calyx slightly longer than peduncle which tapers to large, often saucer-like pedal disc. Up to 14 tentacles (usually 10-12). Lophophore slightly oblique. No sensory papillae in Manx specimens (small papillae occur in other varieties of L.claviformis). Stomach often

Table 15 . Dimensions of *Loxosomella claviformis*. 40 specimens measured. Units are μ

Dimension	Mean	Range
Total length	500	290–686
Calyx length	254	158–370
Peduncle length	246	132–396
Maximum width	181	88–264
Peduncle width	83	62–106

Table 16 Proportions of *Loxosomella claviformis*. Based on 40 specimens

Ratio	Mean	Range
Calyx length:Peduncle length	1·14	·6–2·0
Calyx length:Calyx width	1·44	1·0–2·0
Calyx width:Peduncle width	2·18	1·8–2·8
Peduncle length:Peduncle width	2·93	1·5–5·8

reaches almost to body wall, almost circular; lateral pouches absent. Budding zone opposite top half of stomach. Few buds at a time. Buds directed anteriorly. Longitudinal muscles well-developed in peduncle which has oblique muscles in upper part.

Dimensions from 40 Manx specimens are given in Table 15 and 16

Discussion

Described by Hincks (1880) from a Guernsey Hermione hystrix and later collected on the same host and from Aphrodite aculeata at Plymouth (Atkins 1932) and Roscoff (Bobin and Prenant 1956). Loxosomella claviformis occurred on 8 (44%) of 18 Hermione hystrix and 7 (32%) of 22 Aphrodite aculeata from Manx waters examined. It occurred on 2 of 6 A.aculeata trawled in Luce Bay. Atkins believed the type she called Loxosoma sp. to be restricted to A.aculeata and L.claviformis sensu stricto to be found only on H.hystrix but Bobin and Prenant found both Atkins types and intermediate forms on both A.aculeata and H.hystrix.

L.claviformis occurs on dorsal and ventral surfaces of the hosts as well as on parapodia bases and the ventral surfaces of the élytra.

Embryo have been seen in August, November and February and buds in August and November. L.claviformis has not been collected in other months.

Loxosomella fauveli Bobin and Prenant (Fig I 10)

Description

Calyx oval or pear-shaped in frontal view and joins smoothly with peduncle but in such a way that the two can be readily distinguished. Large, slightly oblique lophophore. 8 tentacles in bud, up to 10-11 in adult. Calyx almost circular in transverse section. No obvious lateral sense-organs, one or two sensory bristles may be present. Stomach wider than long and lacks lateral pouches. Always a small space between stomach and body wall. Buds lateral, level with stomach, rarely more than one or two on each side. Peduncle shorter than calyx, pedal disk slightly developed. Up to 3 embryos

Table 17. Dimensions of *Loxosomella fauveli*. 50 Manx specimens measured (30 from *Hermione hystrix*, 20 from *Aphrodite aculeata*. Data from 72 Plymouth specimens (from *A.aculeata*) from Atkins (1932). Data for Roscoff specimens from Bobin and Prenant (1956). Units are μ.

Source	"Host"	Total length Mean	Total length Range	Calyx length Mean	Calyx length Range	Peduncle length Mean	Peduncle length Range	Calyx width Mean	Calyx width Range	Peduncle width Mean	Peduncle width Range
Isle of Man	*A.aculeata*	346	246–528	234	159–352	112	70–176	149	114–211	58	53–70
	H.hystrix	385	229–572	260	176–370	126	62–202	179	1[illegible]–229	72	44–97
Plymouth	*A.aculeata*	530	180–800	385		225		179			30–80
Roscoff	*A.aculeata* *H.hystrix*.		170–540								

Table 18. Proportions of *Loxosomella fauveli*. Based on 50 specimens collected off the Isle of Man. (30 from *Hermione hystrix* and 20 from *Aphrodite aculeata*.

Ratio	from *H.hystrix* Mean	from *H.hystrix* Range	from *A.aculeata* Mean	from *A.aculeata* Range
Calyx length:Peduncle length	2·11	1·2–3·5	2·15	1·1–2·6
Calyx length: Calyx width	1·44	1·0–1·9	1·54	1·2–2·2
Calyx width:Peduncle width	2·52	2·1–3·4	2·57	1·9–3·6
Peduncle length:Peduncle width	1·8	1·2–2·5	1·95	1·1–3·1

in posterior brood pouch.

Dimensions of L.fauveli are given in Table 17 and 18

Discussion

L.fauveli occurs in large numbers on the parapodia bases, dorsal and ventral body surfaces and the ventral surface of the elytra of Aphrodite aculeata and Hermione hystrix. 9 (50%) of 18 H.hystrix and 9 (41%) of 22 A.aculeata from the Isle of Man and 5 of 6 A.aculeata from Luce Bay bore L.fauveli. L.fauveli often occurs mixed with other entoprocts e.g. L.obesa and L.claviformis. It occurred on 40% of 156 A.aculeata from Plymouth (Atkins 1932).

From Table 17+18 it is obvious that there is little difference between L.fauveli from A.aculeata and those from H.hystrix. Specimens from H.hystrix tend to be a little larger than those from A.aculeata but the proportions of the two groups are almost identical. Both French (Bobin and Prenant 1956) and Manx specimens tend to be smaller than Plymouth specimens.

In Manx collections embryos and buds were present in June, August and November, mature gonads were present in May; neither embryos, buds nor mature gonads were present in specimens collected in February. In the English Channel (Bobin and Prenant 1956) this species is sexually mature and buds from March to October, Atkins (1932) records embryos and mature gonads from April to August and buds in June, August and September.

Loxosomella bouxini Bobin and Prenant (Fig I 11)

Description

Up to 700μ in length. Calyx clearly separated from the thick, cylindrical peduncle. Peduncle swollen in some specimens. Large buds with 8 tentacles, adults usually 10-11. Contour of large lophophore determines that of distal part of calyx. In frontal view calyx either parallel sided or slightly oval. Calyx slightly flattened antero-posteriorly, its width equals about 1½ times its thickness. Very large ovoid stomach usually reaches body wall but there may be a small space between stomach and body wall. Stomach wider than long

Table 19. Dimensions of *Loxosomella bouxini*. 20 specimens measured. Units are μ.

Dimension	Mean	Range
Total length	371	202–616
Peduncle length	200	88–352
Calyx length	172	96–264
Calyx width	162	106–228
Peduncle width	78	44–106

Table 20. Proportions of *Loxosomella bouxini*. Based on 20 specimens.

Ratio	Mean	Range
Calyx length: Peduncle length	1·8	·6–1·5
Calyx length: Calyx width	1·07	·8–1·5
Calyx width: Peduncle width	2·1	1·7–2·8
Peduncle length: Peduncle width	3·2	1·7–3·6

and its superior edge almost tangential to lophophore. Stomach cavity large often filled with debris. Lateral and superior stomach walls very thick, inferior wall thin. Gonads may be so large as to distort lateral walls of calyx. Peduncle $1\frac{1}{2}$ times as long as calyx. Peduncle parenchyma not dense. Up to 2 buds each side, inserted level with inferior part of stomach generally laterally and usually inclined downwards.

Dimensions of 20 Manx specimens are included in Table 19 & 20

Discussion

Previously described only from Concarneau where it occurred on 31 (36%) of 87 Phascolion strombi examined by Bobin and Prenant (1953) L.bouxini occurred in small numbers on 9 (22%) of 41 P.strombi from Manx waters examined. Manx specimens are very similar to French specimens but tend to have a wider range in the proportion calyx length:calyx width. Manx specimens were in mixed populations with L.murmanica and L.nitschei.

Buds were present in April and in material from the "Faunal Collections" labelled "1.8.46". Specimens were not obtained in other months.

Loxosomella cryptae Bobin and Prenant (Fig I 12)

Description

All Manx specimens had 9 short tentacles (Bobin and Prenant found 8-10 tentacles). Calyx slightly compressed antero-posteriorly, almost rectangular in frontal contour, more or less abruptly separated from peduncle. Large lophophore inclined anteriorly. Cuticle thin, uniform, not ridged on peduncle. No enlarged podal disk. Peduncle shorter than calyx in young specimens, longer in large specimens. Stomach a little longer than wide, without lateral pouches, always separated from body wall. Budding zone lateral and opposite centre of stomach. Sensory papillae absent.

The dimensions of the six specimens collected are included in Table 21 and 22

Table 21. Dimensions of *Loxosomella arvyae* from the Isle of Man (6 specimens measured) and from Concarneau (data from Robin and Prenant 1956).

Dimension	Manx specimens	French specimens
Total length	229–370	348–672
Calyx length	141–212	180–312
Peduncle length	88–158	160–360
Calyx width	106–158	120–216
Peduncle width.	44–53	30–65

Table 22. Proportions of *Loxosomella arvyae* from the Isle of Man and from Concarneau (data from Robin and Prenant 1956)

Ratio	Manx		French	
	Mean	Range	Mean	Range
Calyx length:Peduncle length	1·3	1·1–1·6	–	·83–1·25
Calyx length:Calyx width	1·3	1·3–1·4	1·46	1·29–1·66
Calyx width:Peduncle width	2·6	2·4–3·2	3·68	2·70–5·0
Peduncle width:Peduncle length	·41	·34–·5		

Table 23 Dimensions of Loxosomella fungiformis from the Isle of Man (16 specimens measured) and from Concarneau (data from Bobin and Prenant (1953)).

Dimension	Isle of Man		Concarneau	
	Mean	Range	Mean	Range
Total length	192	132–229	–	156–408
Calyx length	118	88–141	–	108–216
Peduncle length	73	44–98	–	48–192
Maximum width	166	114–246	–	132–192
Peduncle width	58	44–62	–	54–90

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Table 24 Proportions of Loxosomella fungiformis from Isle of Man and from Concarneau (data from Bobin and Prenant (1953))

Ratio	Isle of Man		Concarneau	
	Mean	Range	Mean	Range
Calyx length:Peduncle length	1·7	1·2–2·5	1·7	1·2–2·6
Calyx width:Peduncle width	2·9	2·1–4·0	2·3	1·9–3·0
Calyx length:Calyx width	·7	·6–1·2	·9	–
Peduncle length:Peduncle width	1·3	1·0–2·0		

Discussion

Loxosomella aryyae has been collected only once in Manx waters - specimens were found on a Phascolion strombi collected in April. One specimen bore a bud. L.aryyae has previously been recorded only from the type locality, the Bay of Morlaix where it occurred on 3 of 8 P.strombi examined (Bobin and Prenant 1953).

Loxosomella fungiformis Bobin and Prenant (FigI/3)

Description

Maximum length 410 μ. Wide thick calyx joins short thick peduncle almost at right angles giving the animal a fungiform contour. 9-10 (rarely 8) tentacles. Lophophore inclined anteriorly. Calyx often semi-circular in shape, $\frac{2}{3}$ as thick as wide. The much wider than high stomach never reaches body wall, lumen wide but not long, no lateral pouches. Embryos brooded in atrium. Cylindrical peduncle narrows slightly at base into small pedal disk. Cuticle thin on calyx, thicker and ridged on peduncle. Large longitudinal and oblique muscles in peduncle. Some longitudinal fibres extend to lophophore. Dense, opaque, parenchyma. Budding zone lateral at base of stomach, buds horizontal or inclined towards base.

Dimensions of 16 Manx specimens are summarized in Table 23 and 24

Discussion

Described by Bobin and Prenant (1953) from Concarneau where it occurred on 33% of 87 Phascolion strombi examined. L.fungiformis occurred on only 2 of 41 P.strombi from the Isle of Man. L.fungiformis was mixed with L.murmanica and L.nitschei.

Embryos and buds were present in specimens from the "Faunal Collections" dated 1.8.46.

Discussion on hosts and host preferences in the genus Loxosomella.

For the purposes of this discussion the substrates on which Loxosomella spp. have been found in Manx waters can be divided into three groups:

Group A: Worm tubes.

Group B: Polychaeta Aphroditidae. *Hermione hystrix*, *Aphrodite aculeata* and *Lagisca extenuata* often bear species of *Loxosomella*.

Group C: *Phascolion strombi*. 5 *Loxosomella* spp. occur on *P.strombi* in Manx waters.

The three groups will be discussed separately.

A. Worm tubes

Loxosomella marsyops and *Loxosomella* sp.nov. (as well as *Loxosoma* sp.nov.) have been found on the inner, membranous lining of worm tubes. Only one entoproct species occurred in any one worm tube but the ectoproct *Hypophorella expansa* Ehlers occasionally occurs in the same *Chaetopterus* tube as *L.marsyops*. *H.expansa* is colonial and has a thin autozooid-bearing stolon which ramifies among the membranous linings of *Chaetopterus* and *Lanice* tubes. The *H.expansa* zooids protrude their tentacles through small holes into the lumen of the tube. *L.marsyops* is solitary and feeds by ciliated tentacles in the lumen of the worm tube. But despite the similarities in habit and habitat competition is probably reduced by the fact that while *H.expansa* occurs most often in the newer parts of the tube *L.marsyops* is most often present in the older more friable parts of the tube. *H.expansa* occurred in about 80% of the *Chaetopterus* tubes examined, *L.marsyops* occurred rarely.

B. Polychaeta Aphroditidae

Loxosomella compressa has been found only on the notopodial setae of *Lagisca extenuata*.

Loxosomella claviformis and *L.fauveli* occur on both *Aphrodite aculeata* and *Hermione hystrix*, *L.obesa* occurs on *A.aculeata*.

Atkins (1932) examined 146 *A.aculeata* and a few *H.hystrix* at Plymouth. Her results can be summarized as follows: *L.obesum* occurred on the dorsal surface of 12.3% of the *A.aculeata*. *L.fauveli* occurred on 39% of the *A.aculeata* and was not frequently present on the ventral surface but when present on the dorsal surface was in greater numbers than on the ventral surface, it was absent from *H.hystrix*.

Table 25 Distribution of *Loxosomella claviformis*, *L. obesum* and *L. fauveli* on *Aphrodite aculeata* and *Hermione hystrix*. Plymouth data from Atkins (1932), Roscoff data from Robin and Prenant (1953). In the table the bracketed figure is the number of worms examined. + a few specimens occur, ++ specimens not uncommon, +++ most specimens (or all specimens) occur on this surface.

	Loxosomella obesum				*Loxosomella claviformis*				*Loxosomella fauveli*			
		Abundance on				Abundance on				Abundance on		
	% Occurrence	Dorsal surface	Ventral surface	Elytrae	% Occurrence	Dorsal surface	Ventral surface	Elytrae	% Occurrence	Dorsal surface	Ventral surface	Elytrae
A. aculeata from:												
Plymouth (146)	12 3	+++	-	-	3	+	-	-	39	++	+++	+
Roscoff (9)	-	-	-	-	33	+	+++	-	11	+	+++	+
Port Erin (22)	32	+++	-	+	27	+	+++	+	43	++	+	+
H. hystrix from												
Plymouth (few)	-	-	-	-	Most	++	+++	+	-	-	-	-
Roscoff (19)	-	-	-	-	Most	+	+++	+	Most	+++	+	++
Port Erin (18)	-	-	+	-	42	-	+++	+	50	+++	+	+

Typical L.claviformis were found only on H.hystrix and occurred all over the ventral surface and on the parapodia, when present dorsally they were in greater numbers on the dorsal surface than on the ventral surface of the elytrae. On one A.aculeata Atkins found "Loxosoma sp." which Bobin and Prenant later (1956) showed to be a variety of L.claviformis. Atkins (1932) remarked "there seems to be no reason why L.claviformis should not be found on A.aculeata, or L.fauveli on H.hystrix".

Bobin and Prenant (1953), working at Roscoff, examined 9 A.aculeata and 19 H.hystrix. They did not find L.obesa. L.claviformis occurred on both worms; it was found most frequently and in largest numbers on the ventral surfaces, some occurred on the dorsal surface but few on the elytrae. L.fauveli occurred regularly under the elytrae and on the dorsal surface of H.hystrix, a few were found on the ventral surface of one H.hystrix, and it was more abundant on the ventral than the dorsal surface of the one A.aculeata on which it was found. Examples of L.fagei Bobin and Prenant and Loxosoma loricatum Harmer were also found on Roscoff H.hystrix. At Port Erin during 1959-62 I examined 22 A.aculeata and 18 H.hystrix.

L.obesa occurred on the posterior dorsal surface and elytrae of 31% of the A.aculeata. L.fauveli occurred on 43% A.aculeata and 50% H.hystrix on both it is most frequently present on the dorsal surface but is occasionally present in smaller numbers on the ventral body surface and ventral surface of the elytrae. (cf. with situation at Plymouth (above and Table 25)). L.claviformis (Port Erin specimens are L.claviformis var Bobin and Prenant = Loxosoma sp. Atkins) occurred on 27% of A.aculeata and 44% H.hystrix. On Hermione it was almost exclusively present on the ventral surface (only 1 individual was found on the elytrae); on Aphrodite it was most frequent on the ventral surface but some occurred on the dorsal surface and on the ventral surface of the elytrae. This is similar to the situation at Roscoff but differs from that at Plymouth (see above and Table 25).

The data from Plymouth, Roscoff and Port Erin are summarized

Table 26 Percentage occurrence of Entoprocts on *Phascolion strombi* from Roscoff, Concarneau and Port Erin. French data from Bobin and Prenant (1953).

Entoproct species	Locality and percentage occurrence of Entoprocts			
	Port Erin *P. strombi* (26) from *Turritella*	Port Erin *P. strombi* (15) from *Dentalium*	Concarneau (87)	Roscoff (8)
Loxosomella murmanica	42	13	65	100
Loxosomella nitschei	73	53	55	-
Loxosomella arvyca	-	6·6	-	37·5
Loxosomella cuenoti	-	-	16	-
Loxosomella fungiformis	7.6	-	30	-
Loxosomella bouxini	35	6.6	36	-
Loxosomella atkinsi	-	-	-	12·5

in Table 25. It can be said as a general conclusion that although mixed populations occur on both A.aculeata and H.hystrix the entoprocts involved show slight differences in distribution on the hosts which will tend to reduce inter-specific competition. L.claviformis and L.fauveli occur on both A.aculeata and H.hystrix. The two worms have slightly different distributions H.hystrix being most common on gravelly grouhds and A.aculeata on sandy grounds. Thus by infecting two 'hosts' the entoprocts gain a wider distribution.

C. Phascolion strombi (Montagu)

Specimens of P.strombi were collected off the south of the Isle of Man from depths between 20 and 40f. (37-73 m.). P.strombi occurs in dead Turritella communis Risso, Aporrhais pes-pelecani da Costa and Dentalium entalis L. shells. 5 Entroproct species L.murmanica, L.nitschei, L.bouxini, L.arvyae and L.fungiformis were fouhd on the sipunculids. Mixed populations of up to 3 species occurred on single P.strombi. L.atkinsae Bobin and Prenant, L.cuenoti Bobin and Prenant and L.brumpti Nilus have been recorded from P.strombi elsewhere but not from the Isle of Man. Bobin and Prenant (1953 a, b, c,) examined Phascolion strombi from Roscoff, Morlaix, Concarneau, Dinard, etc. and their findings together with those from Port Erin are summarized in Table 26. Most of the French Phascolion were from Turritella shells; the data from Manx P.strombi from Dentalium shells and from Turritella (+ 1 from A.pes-pelecani) are presented separately.

From Table 26 it is apparent that the % occurrence of all the Loxosomella spp. except L.nitschei is lowest at Port Erin. At Port Erin L.nitschei occurs more frequently than L.murmanica; the opposite is true at Concarneau and L.nitschei was not recorded at Roscoff. All entoprocts at Port Erin occur less frequently on P.strombi from Dentalium shells than on those from Turritella shells. This may reflect the difference in shape of Turritella and Dentalium shells.

Bobin and Prenant (1953) state that the distribution of Entoprocts on P.strombi does not follow any pattern but they note the abundance of Loxosomella, particularly L.nitschei and L.murmanica, on the

TEXT BOUND INTO

THE SPINE

Table 27 Distribution of *Loxosomella* species on *Phascolion strombi* from off the Isle of Man. (See text p. 22 for explanation of division of *P. strombi*.)

Entoprost species	Number of occurrences (26 possible)			Number of specimens			
	Around anus	Spiny region	Smooth region	Around anus	Spiny region	Smooth region	Tot
Loxosomella nitschei	13	15	8	72	110	22	2
Loxosomella murmanica	7	10	-	16	114	-	1
Loxosomella bouxini	5	6	1	33	18	7	
Loxosomella fungiformis	-	1	2	-	[illegible]	10	
Loxosomella [illegible]	-	1	-	-	9	-	
Total				121	259	39	5

convex posterior flank of P. strombi. During the present investigation the position of each Loxosomella on the P. strombi was noted. For the purposes of the investigation each Phascolion was considered to be divisible into three regions (a) the area around the anus, (b) the posterior region with papillae and cuticular spines and hooks ("the spiny region") and (c) the anterior "smooth" region without cuticular hooks or spines. Region (a) is much smaller in area than (b) or (c) and (b) is smaller than (c). The number of each Loxosomella species in each region of the Phascolion are given in Table 27. Although the numbers are small some interesting points emerge. There are many more entoprocts present around the anus and on the "spiny" region than on the "smooth" part. Some species appear to show preferences for particular sites e.g. L. nitschei is most common around the anus and on the "spiny" part, L. murmanica is absent from the "smooth" region and abundant in the "spiny" area, L. bouxini is commonest around the anus, L. fungiformis and L. aryyae have not been found around the anus.

L. murmanica and L. nitschei are rarely present together on a Phascolion but whichever is present is usually dominant. Bobin and Prenant found this situation in French populations also.

FAMILY PEDICELLINIDAE

Diagnosis

Marine or brackish. Sessile, colonial Entoprocts with the calyx separated from the peduncle by a diaphragm. Calyx deciduous. Larva settles and forms oözooid which developes a peduncle and calyx, peduncle then gives off stolon which usually adheres to substrate. Stolons bud off peduncles which develop calyces, buds do not detach but form colonies.

Manx genera are Pedicellina M. Sars and Barentsia Hincks.

GENUS PEDICELLINA M. Sars

Diagnosis

Cylindrical peduncle is muscular and flexible throughout. No basal muscular node. Two Manx species Pedicellina cernua (Pallas) and P.nutans Dalyell.

Key to the Manx species of Pedicellina

(After Bobin and Prenant 1956)

Peduncle not or very slightly tapered, not narrowing markedly below calyx which it joins by a large surface. Large calyx, up to 6 times peduncle width. Dorsal side of calyx is convex
Pedicellina cernua

More of less fusiform peduncle retracted almost to a point below calyx. Calyx small, not more than 3 times width of peduncle. Asymmetrical in profile
P.nutans

Several forms of P.cernua have been described, some were originally desiganted species but recent work (and the present study) shows that more than one type of zooid may be budded off a single stolon Joliet (1877) distinguished three forms:

f.glabra	P.glabra of Dalyell (1848) and Ehlërs (1890) peduncle and calyx glabrous.
f.echinata	P.hirsutum Ehllers 1890 and P.echinatum M.Sars 1835 all zooids with spines on peduncle and calyx.
f.typica	Peduncle spiny, calyx glabrous.

F.typica is most common in Manx waters, the other forms are rare.

Pedicellina cernua (Pallas) (Fig I 14)

The largest Entoproct collected, zooids are easily visible to the unaided eye and vary in length from 0.4 - 5 mm. It occurred in most localities from between L.W.S.T.L. and 50 f. (91 m.) and seems to occur wherever there are suitable supports. Its distribution on various supports is discussed on p. 14 in comparison with Barentsia gracilis. Colonies are largest in summer and it is then (July - October) that reproduction takes place. Small colonies of 1 or 2 zooids have been found during the winter months, colonies collected in winter are often without calyces. In the Channel P.cernua reproduces all summer (Bobin and Prenant 1956); Lo Bianco (1908) found reproducing colonies at Naples in November.

P.cernua is widely distributed in the Arctic, on N.Atlantic coasts and in the Mediterranean.

Pedicellina nutans Dalyell. (See Bobin and Prenant 1956)

Collected only twice, once from L.W.S.T.L. at Port Erin Breakwater and once on Bugula plumosa (Pallas) from 18 f. (33 m.) off Port Erin Breakwater.

P.nutans is recorded from several points on British, Norwegian and Danish coasts and from the St. Lawrence Estuary.

GENUS BARENTSIA Hincks

Diagnosis

Peduncle not muscular throughout its length but has muscular regions separated by narrower, non-muscular rigid regions with strong cuticle.

Two Manx species Barentsia gracilis and Barentsia sp.nov?

Barentsia gracilis (M.Sars)

Smaller than P.cernua zooids range from 0.5 - 1.5 mm. and calyx from 100 - 350 μ . Easily recognised by its characteristic peduncle consisting of basal muscular node and narrow rigid stalk which may have one or more small muscular nodes along its length. 12-14 tentacles.

Widely distributed from L.W.S.T.L. to 45 f. (82 m.) where it occurred on a Nephrops norvegicus L. carapace. The stiff puppet-like movements of the peduncle in living material attract attention. B.gracilis occurs on a wide variety of supports (shells, stones, hydroids, ascidians and even Hermione hystrix). A discussion on its support preferences is given below.

Most Manx specimens are of the typical type but occasional specimens of var.nodosa with up to 3 muscular articulations on the peduncle have been collected. Some colonies have both typical and nodosa zooids.

Barentsia sp.nov? (Fig 15+16)

Specimens of a colonial entoproct which differs from other described species (See Marcus 1949) have been collected on several

Table 2: Dimensions of *Barentsia* sp nov? 20 specimens measured. Units are μ

Dimension	Mean	Range
Total length	1090	707–1400
Calyx length	143	122–272
Calyx width	110	68–176
Total peduncle length	670	580–1280
Rigid part of peduncle length	280	136–612
Basal muscular node length	117	109–310
Basal muscular node width	87	68–136

occasions from the walls of the Aquarium tanks at Port Erin Marine Biological Station.

Description (See Fig I 15 and Table 28)

Stolon, 30 - 40 μ thick, attached to substrate, bears zooids singly in small groups up to 1 mm. apart. Zooids up to 1.5 mm. total length. Calyx small (mean length 140 μ) and slightly assymetrical in lateral view. 10-11 tentacles. Above its basal muscular node the peduncle is divisible into two more or less distinct regions. The proximal part varies in length from a few μ to about half the peduncle length and is narrow, rigid and non-muscular. The distal part is thicker, membranous walled, very flexible and muscular. The distal part of the peduncle narrows sharply at its junction with calyx. Spines are absent from the peduncle and the calyx.

In life the zooid can be moved by the muscular base of the peduncle and the distal part of the peduncle is also highly mobile and readily takes up a ∩ shape (See Fig I 16).

Affinities

Other Barentsia species in which the upper part of the stalk is flexible are B.major Hincks, B.laxa Kirkpatrick, and B.stiria Jullien and Calvet. Each is listed below together with some characters by which it differs from Barentsia sp.nov?

B.major	Lower part of stalk annulated. Tentacles numerous (Hincks 1888) Calyx very large.
B.laxa	Up to 9 mm. Fleshy part of stalk annulated. 13-23 tentacles.
B.stiria	Fleshy part of stalk only ½ calyx length. 12 tentacles.

The Distribution of Pedicellina cernua and Barentsia gracilis on different supports.

P.cernua and B.gracilis occur on a wide variety of supports; specimens of both species have been collected on most types of shell, on stones, hydroids, ectoprocts, spider crabs, lobsters, etc.

Table 29. Distribution of *Pedicellina cernua* and *Barentsia gracilis* on various supports. The six supports on which each species is most common are listed together with the percentage occurence of the species on each support. 141 *P.cernua* and 245 *B.gracilis* colonies were examined.

P.cernua Support	% of Total on each support	*B.gracilis*	% of Total on each support
1. Hydroid	31	1. *Chlamys opercularis* outer surface of dead shell	30·6
2. *Chlamys opercularis* outer surface of dead shell	16	2. Living *Chlamys opercularis*	13·0
3. Living *Chlamys opercularis*	15	3. *Chlamys opercularis* inner surface of dead shell	9·7
4. *Modiolus modiolus* dead shell	5·6	4. *Pecten maximus* outer surface of dead shell.	7·7
5. *Eucratea loricata*	4·2	5. Hydroid	7·3
6. *Pecten Maximus* outer surface of dead shell.	3·5	6. *Modiolus modiolus*	5·3
Etc.	24·7	Etc.	26·0
Zoophyte	42·9	Zoophyte	11·3
Inner surface of dead shell	3·2	Inner surface of dead shell	21·2
Outer surface of dead shell	33·3	Outer surface of dead shell	49·0
Total on dead shell	36·5	Total on dead shell	70·2

B.gracilis has been observed on tiny Anomia spat, on small Emarginula reticulata Forbes and Hanley, on the operculae of Pomatoceros triqueter (L.), on the legs of pycnogonids, on barnacle valves, on spines of Hermione hystrix and on the ventral surface of Aphrodite aculeata; P.cernua rarely occurs on such bizarre supports. P.cernua is less common than B.gracilis; 224 P.cernua were noted in a series of samples which contained 377 B.gracilis colonies. Although the two species occur on a similar range of supports the quantitative distribution of the two species on each type of support differs considerably (See Table 29). 42% of P.cernua colonies occur on erect zoophytes but only 11.3% of B.gracilis colonies grow on these supports. B.gracilis is about half as common on inner shell surfaces as outer while P.cernua is 10 times more abundant on outer than on inner shell surfaces. Live Chlamys opercularis are colonised by both P.cernua and B.gracilis. In one sample of 90 C.opercularis 56 bore B.gracilis and 50 P.cernua, 21 of a sample of 33 lower valves carried B.gracilis and 9 P.cernua and of 21 upper valves in another sample 8 bore B.gracilis and 5 P.cernua. On these shells P.cernua is usually confined to the ears and around the margin of the shell while B.gracilis occurs all over the shell surface.

Thus although these two species occur on similar supports their distribution on the various supports differ.

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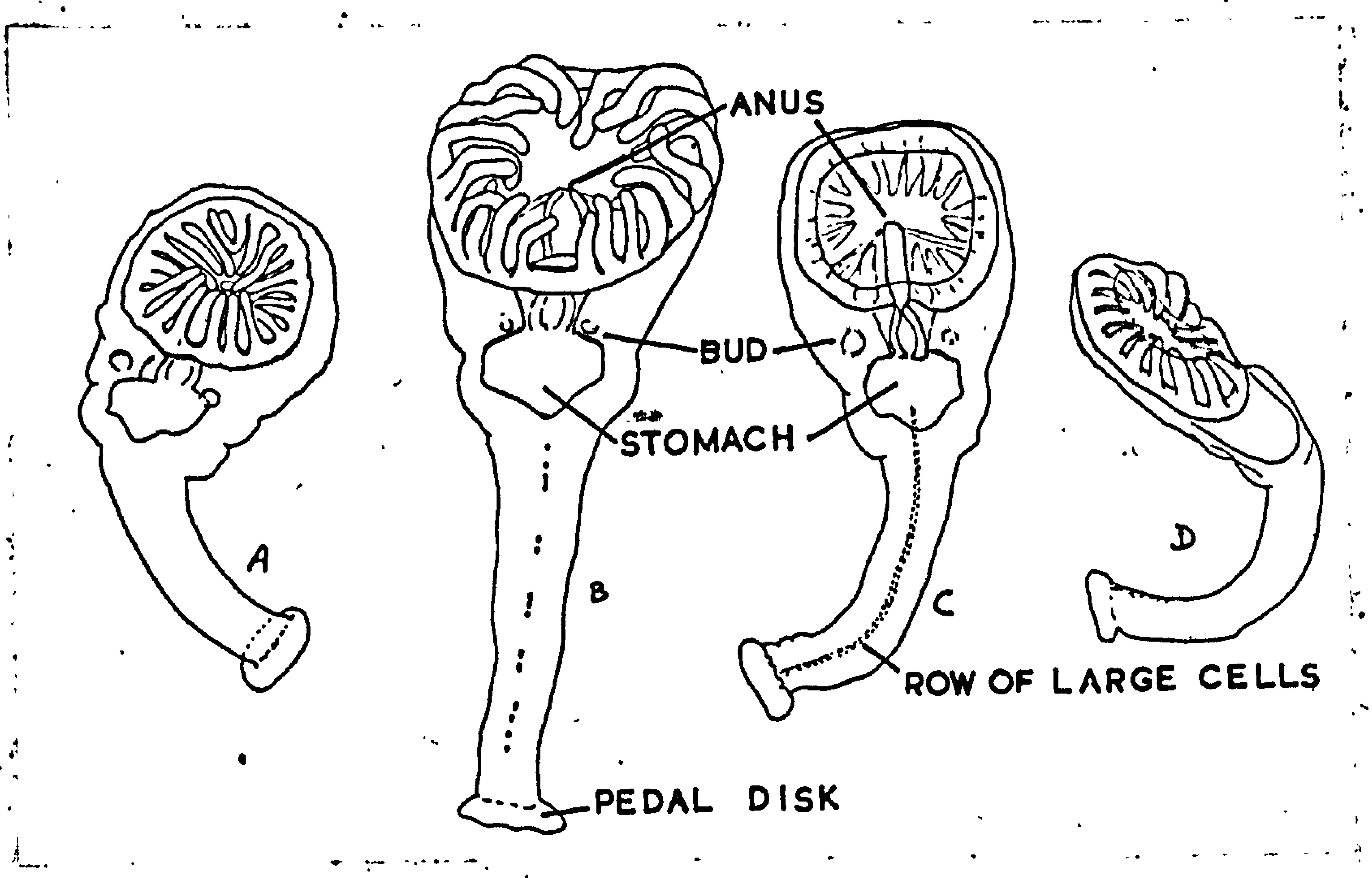

Fig. I.i. Loxosoma sp. nov. A.Frontal view of contracted specimen (Length 853 μ). B. Frontal view of partly contracted specimen (Length 1188) C. Posterior view of partly contracted specimen (Length 924 μ) D. Lateral view of contracted specimen (Length 915 μ). Tiny buds can be seen in specimens A, B and C. Specimens are not drawn to the same scale.

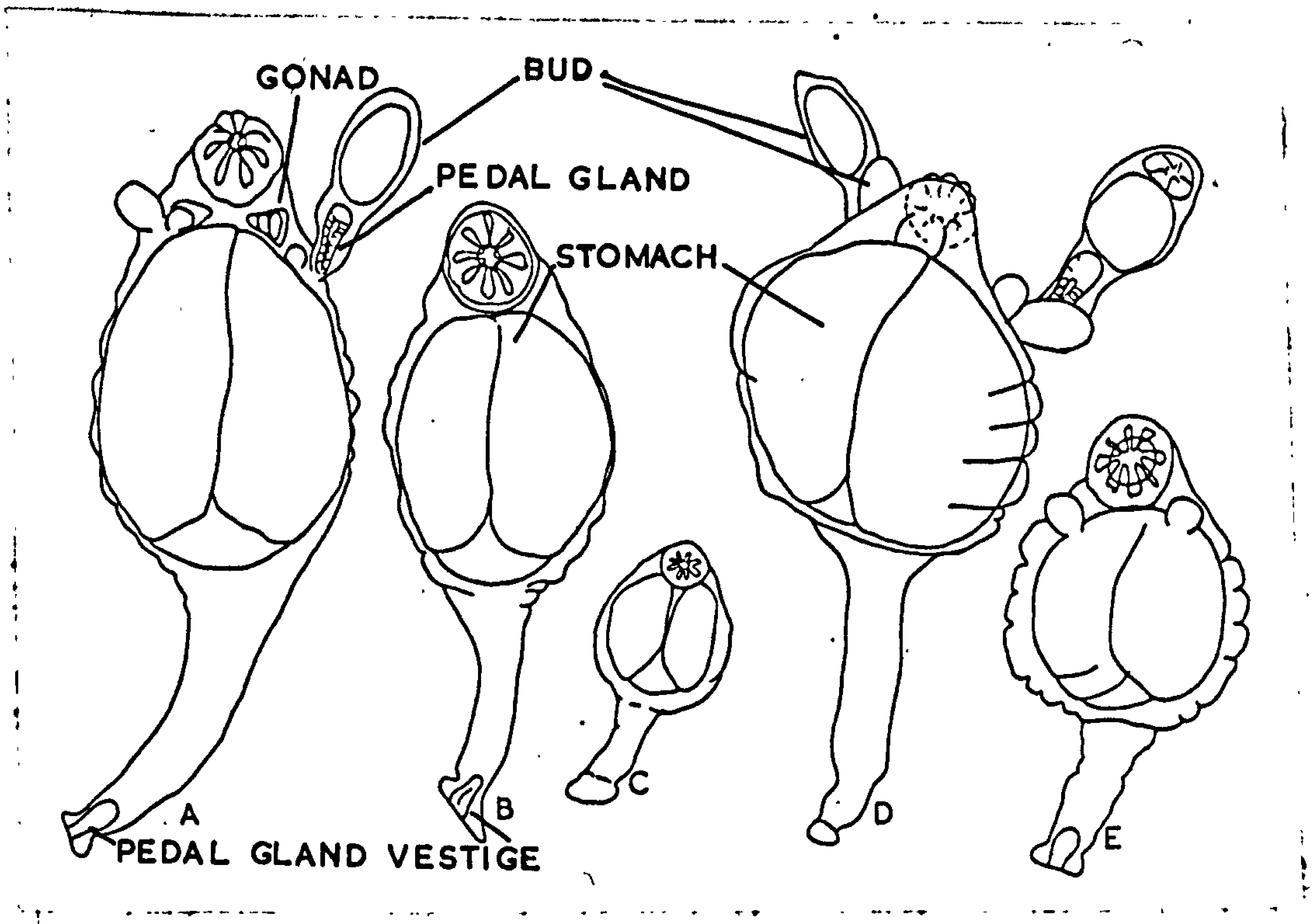

Fig. I. 2. Loxosomella obesa. A. Frontal view of Type 2 specimen (Length 1364 μ). Female gonads and four developing buds are visible. B. Frontal view of Type 2 specimen (Length 1056 μ). C. Frontal view of small Type 3 specimen (Length 563 μ). D. Posterior view of large Type 3 specimen (Length 1249 μ). Five buds are partially visible. E. Frontal view of Type 3 specimen (Length 862 μ). Two buds are visible. Vestiges of the pedal gland are visible in specimens A, B and E. All specimens are not drawn to the same scale.

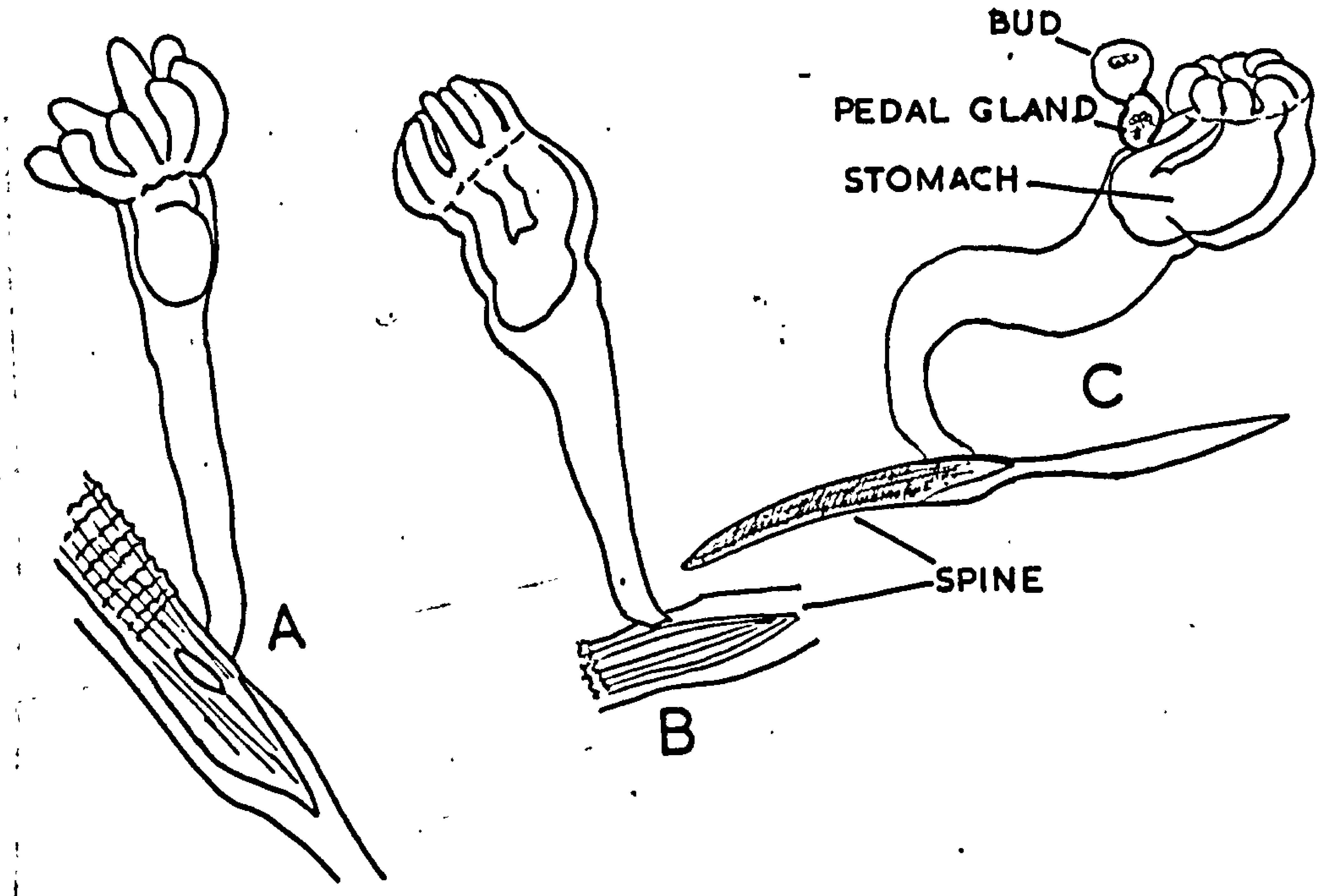

Fig. I.3. *Loxosomella compressa*. Specimens on notopodial spines of *Lagisca extenuata*. A. Frontal view of expanded specimen (Length 598 μ). B. Lateral view of partly contracted specimen (Length 616 μ). C. Lateral view of contracted specimen(Length 651 μ) with one bud. Specimens are not drawn to the same scale.

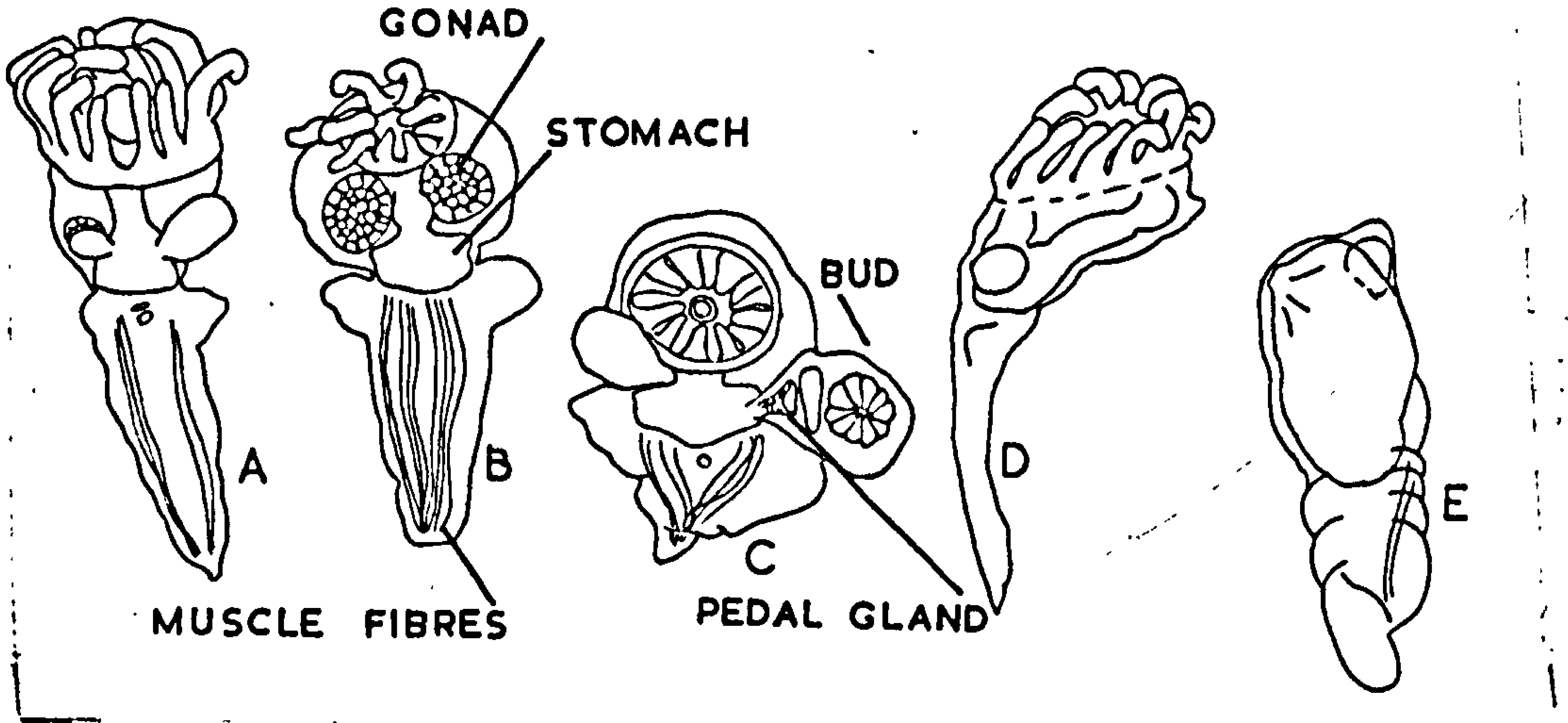

Fig. I.4. *Loxosomella marsyops*. A. Frontal view of fully extended specimen (Length 730 μ). One gonad and two buds are visible. B. Frontal view of partly contracted specimen (Length 633 μ) Two gonads are visible. C. Fully contracted specimen (Length 422 μ) with two buds. D. Lateral view of A. E. Lateral view of fully contracted specimen (Length 448 μ) . Specimens are not drawn to the same scale.

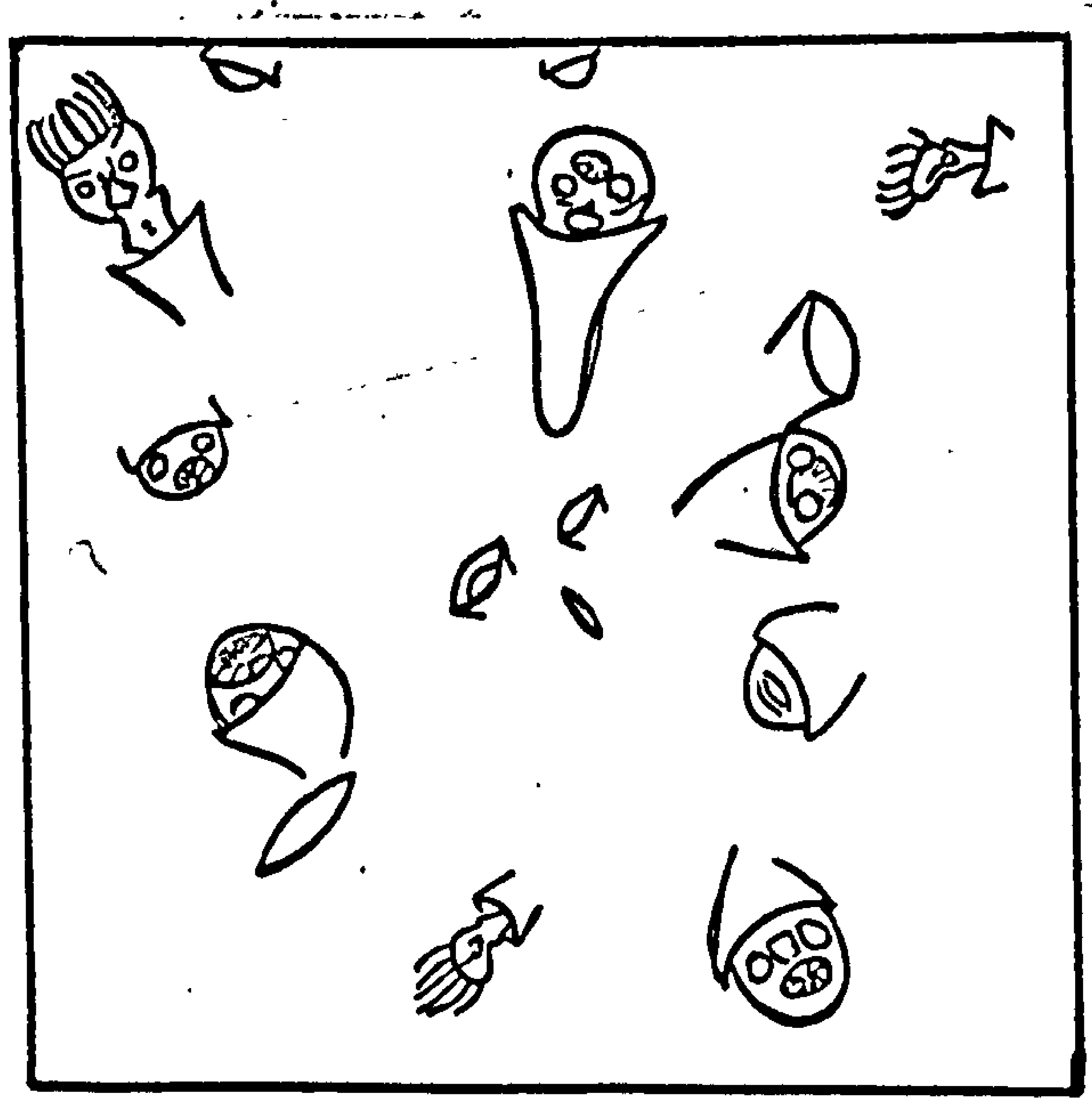

Fig. I.5. Semi-diagrammatic representation of a dense Loxosomella marsyops 'colony' in a Chaetopterus tube. The area included is 2·5 mm²

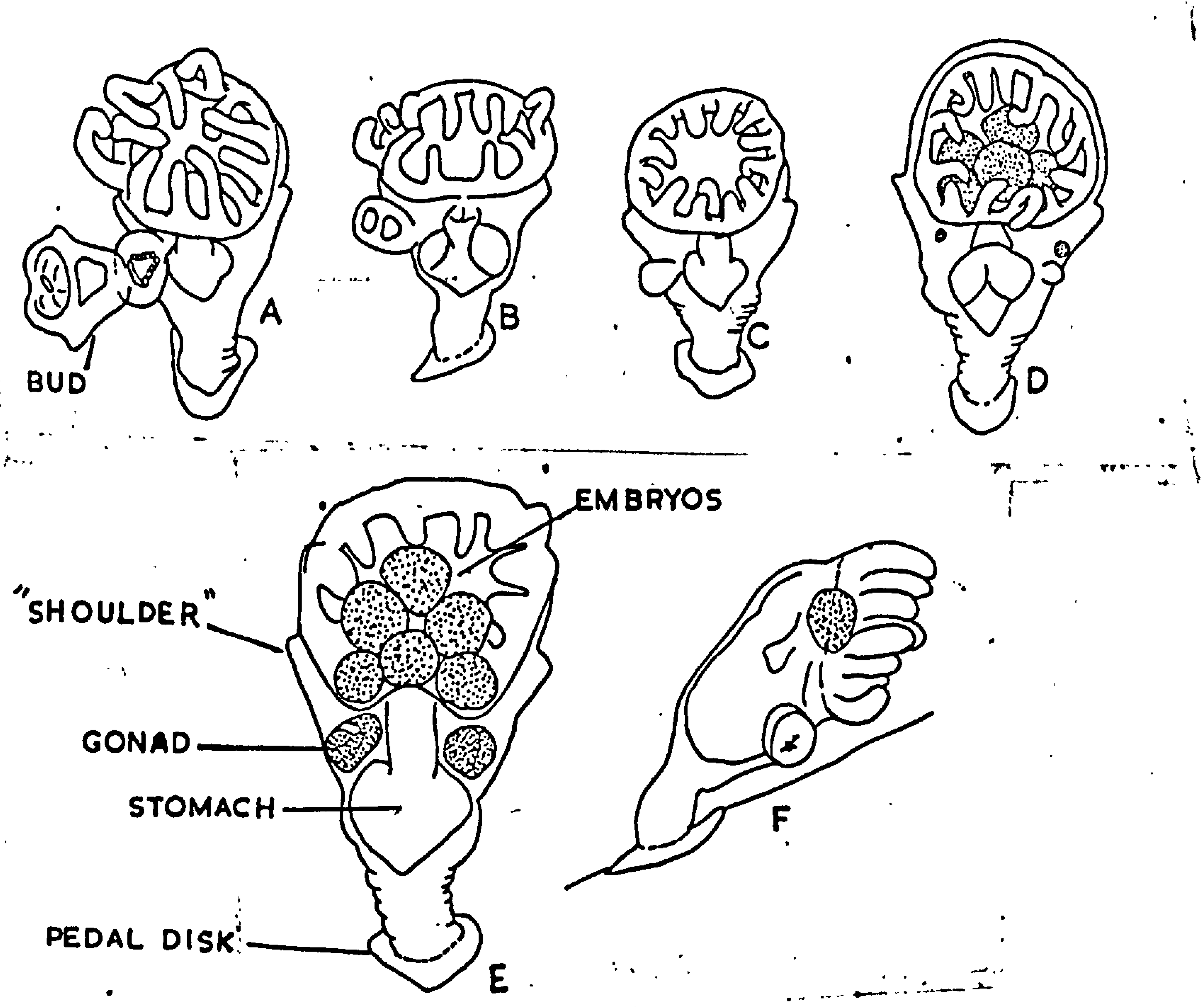

Fig. I. 6. Loxosomella sp.nov. A. Frontal view of expanded specimen (Length 343 μ) with one bud. B. Frontal view of partly contracted specimen (Length 281 μ) with one bud. C. Frontal view of partly contracted specimen (Length 281 μ) with one bud. D. Frontal view of partly contracted specimen (Length 440 μ) with two small gonads, five embryos and one bud. E. Posterior view of specimen (Length 413 μ) with two gonads and six embryos. F. Lateral view of specimen (Length 343 μ), one embryo and one bud are visible. The specimens are not drawn to the same scale.

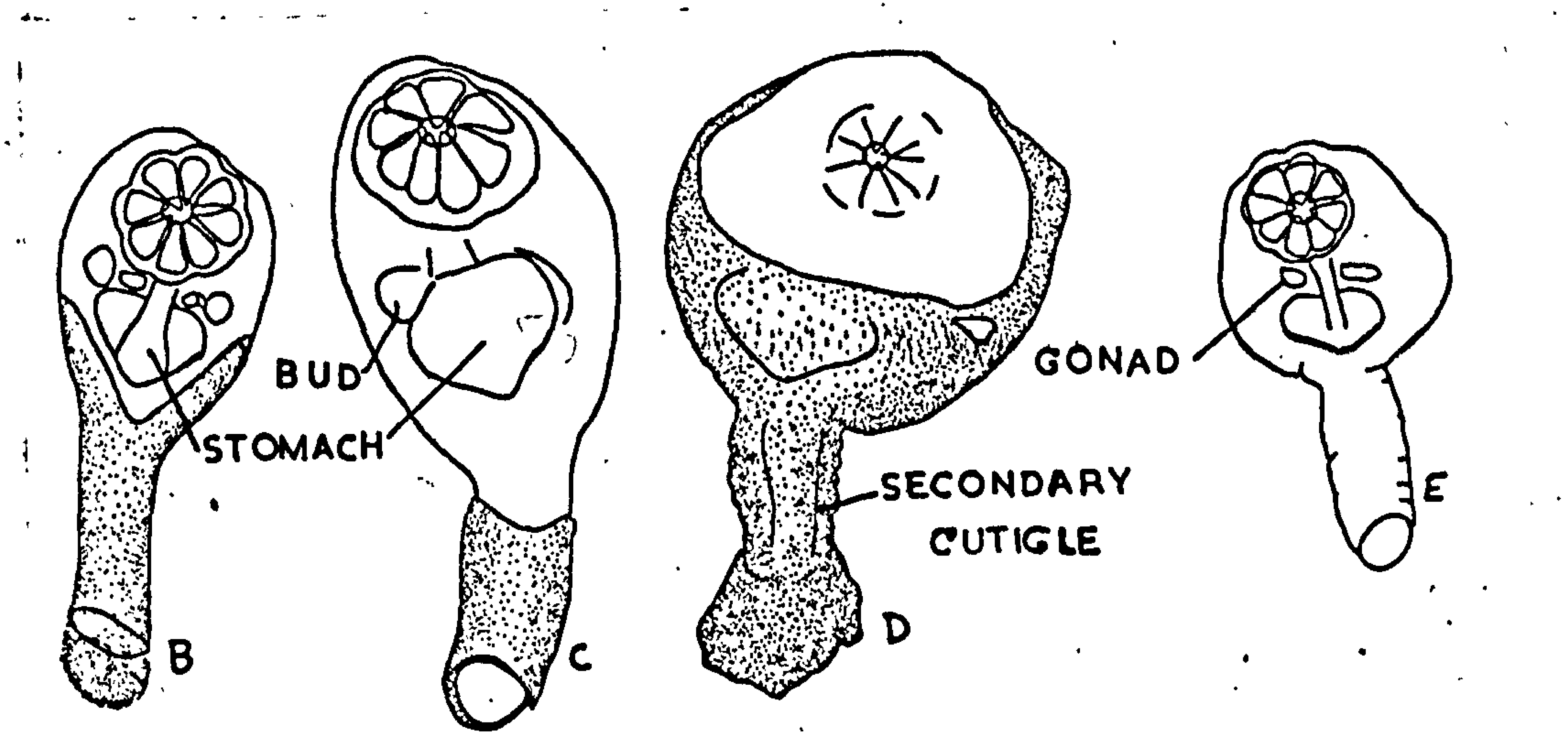

Fig. I. 7. Loxosomella murmanica. All specimens are in frontal view. B. Contracted specimen (Length 326 μ) partly encased in 'secondary cuticle'. C. Contracted specimen (Length 396 μ) partly encased in 'secondary cuticle'. D. Specimen (Length 440 μ) almost completely encased in 'secondary cuticle'. E. Specimen (Length 264 μ) without 'secondary cuticle'. Specimens are not drawn to the same scale.

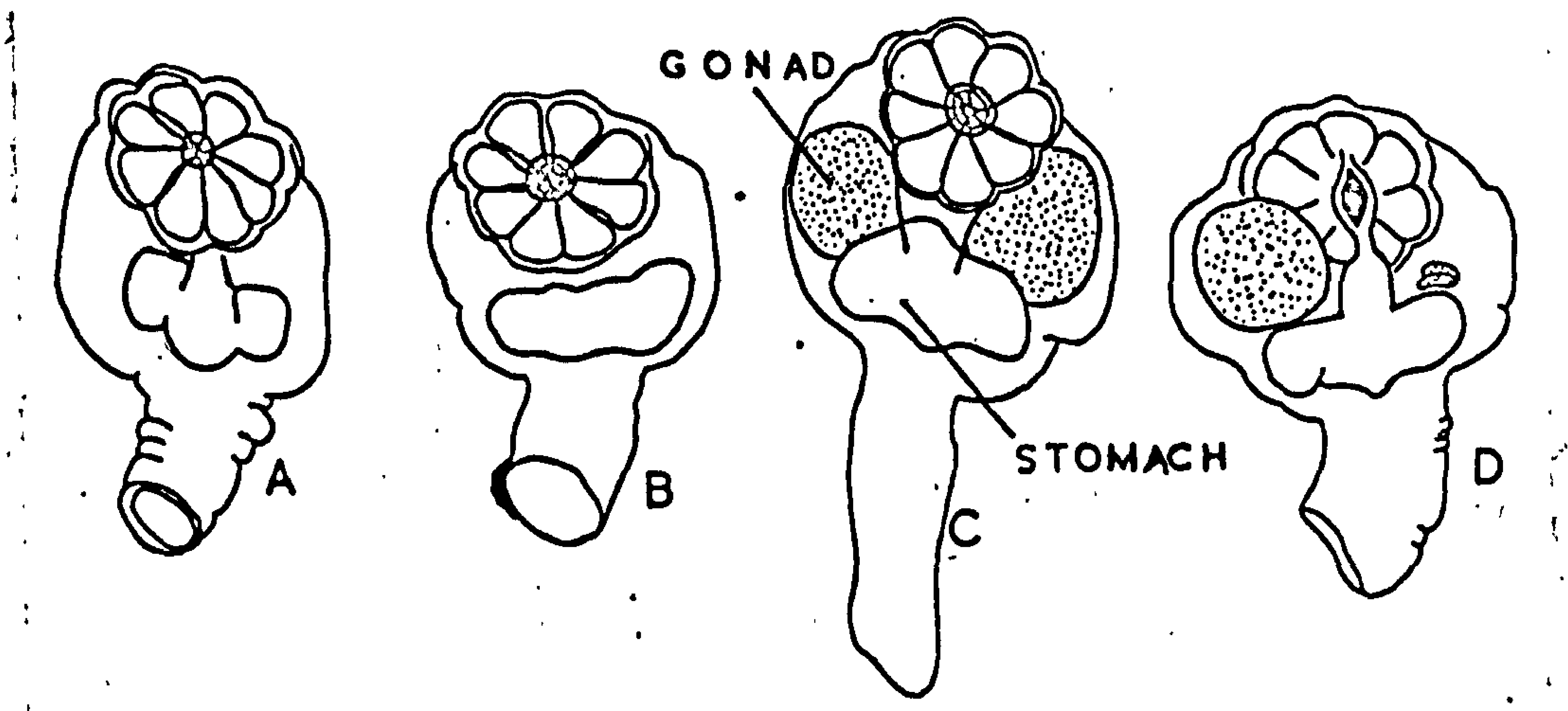

Fig. I. 8. Loxosomella nitschei. A. Frontal view of contracted specimen (Length 264 μ) B. Frontal view of contracted specimen (Length 264 μ) C. Frontal view of contracted specimen (Length 334 μ) with two large gonads. D. Frontal view of contracted specimen (Length 308 μ) with two gonads. Specimens are not drawn to the same scale.

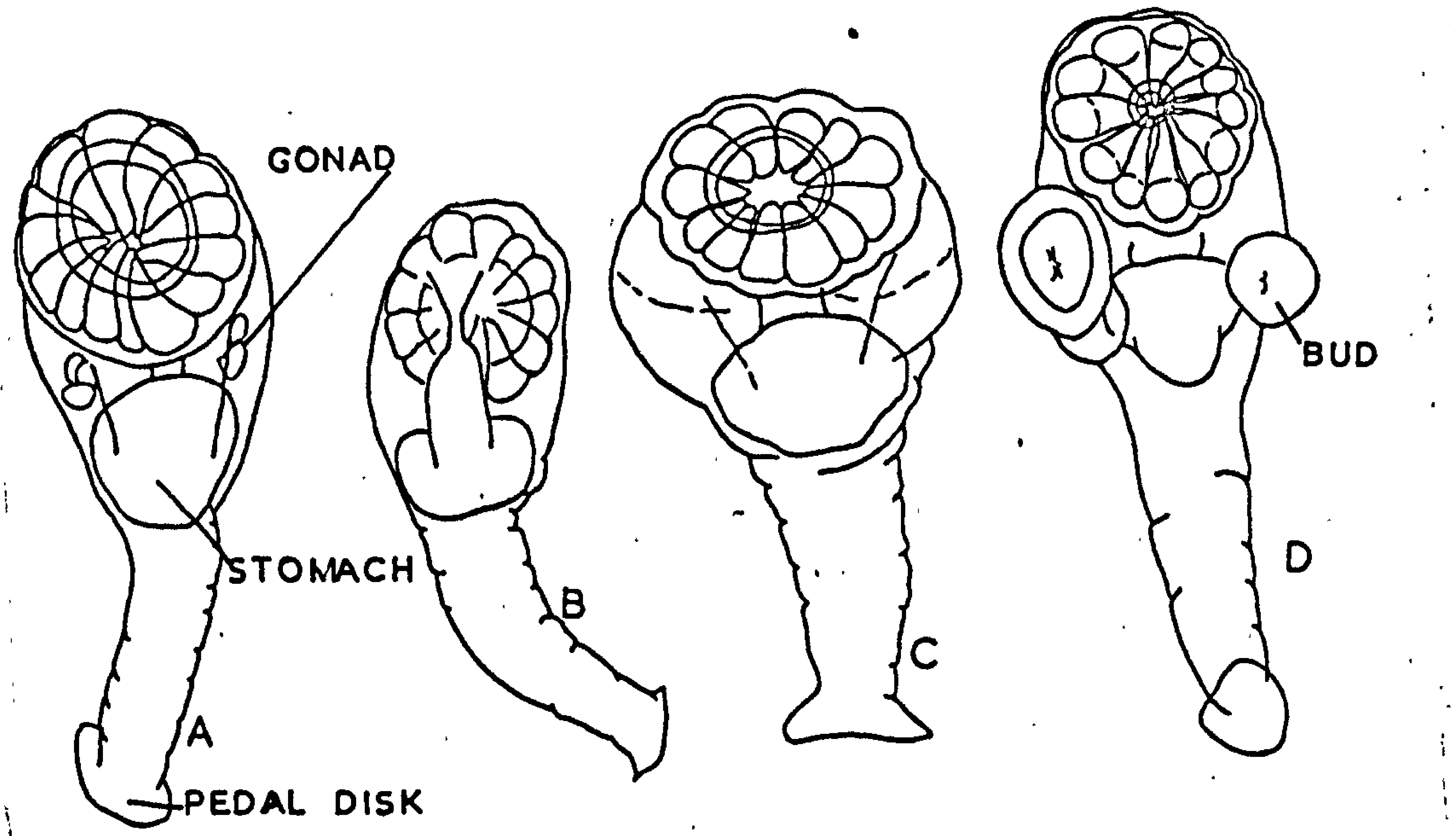

Fig. I. 9. Loxosomella claviformis. A. Frontal view of contracted specimen (Length 528 μ) with two small gonads. B. Posterior view of specimen (Length 431 μ). C. Frontal view of specimen (Length 545 μ). D. Frontal view of specimen (Length 563 μ) with two buds. Specimens are not drawn to the same scale.

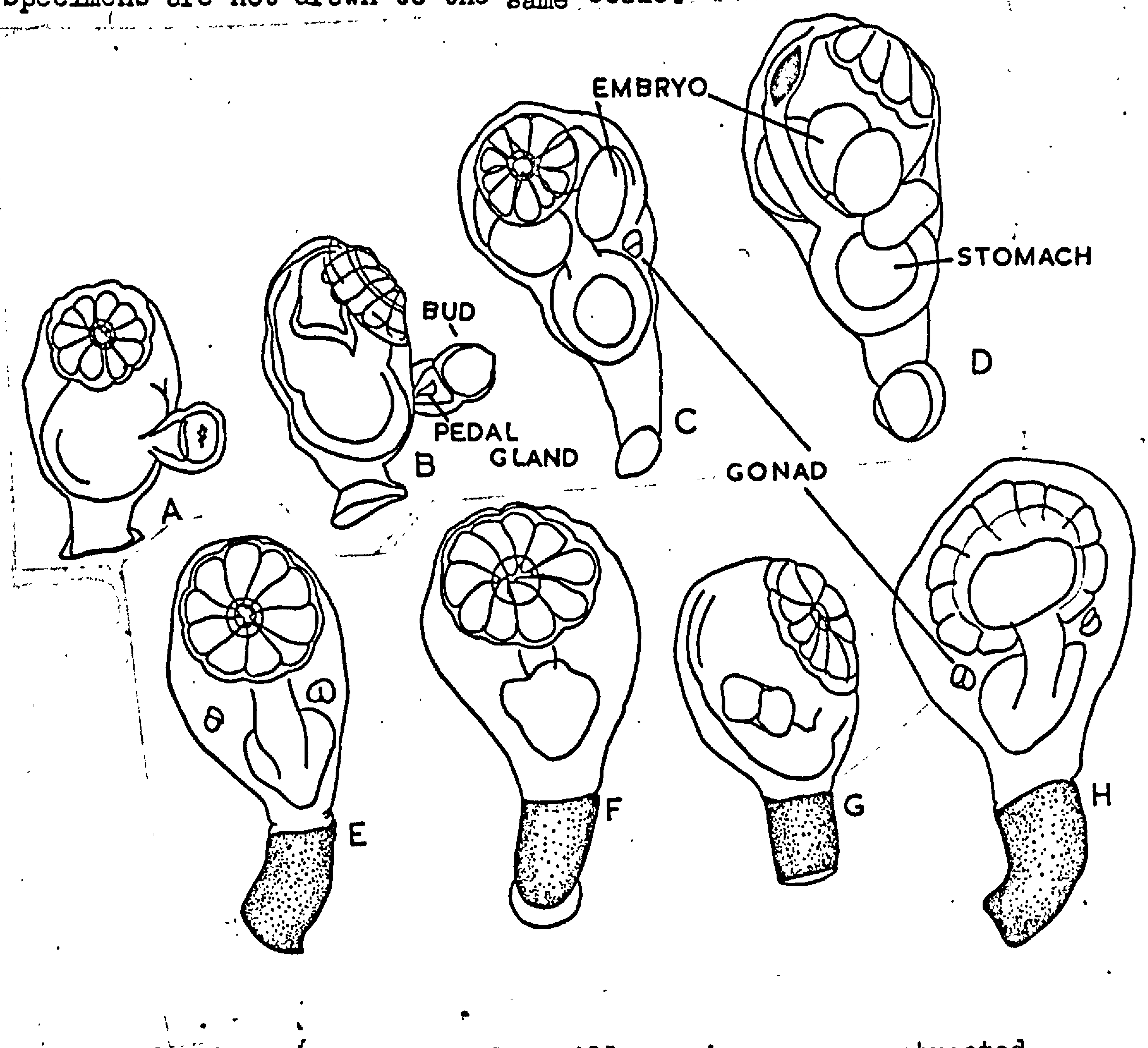

Fig. I.10. Loxosomella fauveli. All specimens are contracted. A. Frontal view of specimen (Length 325 μ) with one bud. B. Lateral view of A. C. Frontal view of specimen (Length 458 μ) with two embryos and one gonad visible. D. Postero-lateral view of specimen (Length 484 μ) with four embryos visible. E. Frontal view of specimen (Length 387 μ) with two small gonads. F. Frontal view of specimen (Length 502 μ). G. Lateral view of specimen (Length 352 μ) with female gonad. H. Posterior view of specimen (Length 484 μ) with gonads visible. The peduncles of specimens E-H are partly encased in a brown substance. Specimens are not drawn to the same scale

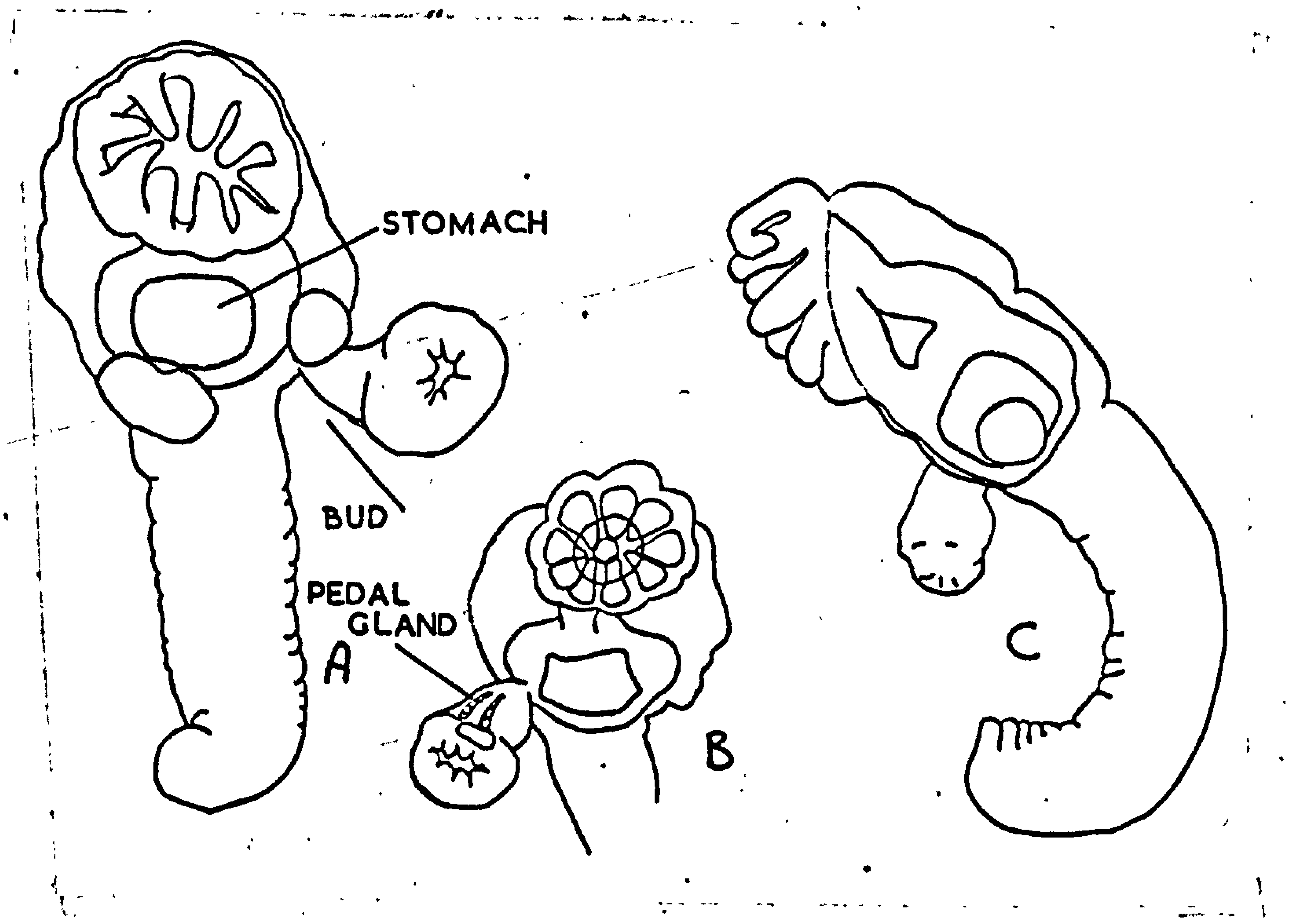

Fig. I. 11. Loxosomella bouxini. A. Frontal view of specimen (Length 475 μ) with three buds. B. Frontal view of calyx of a damaged specimen. One bud is present. C. Lateral view of specimen (length 616 μ) with buds.

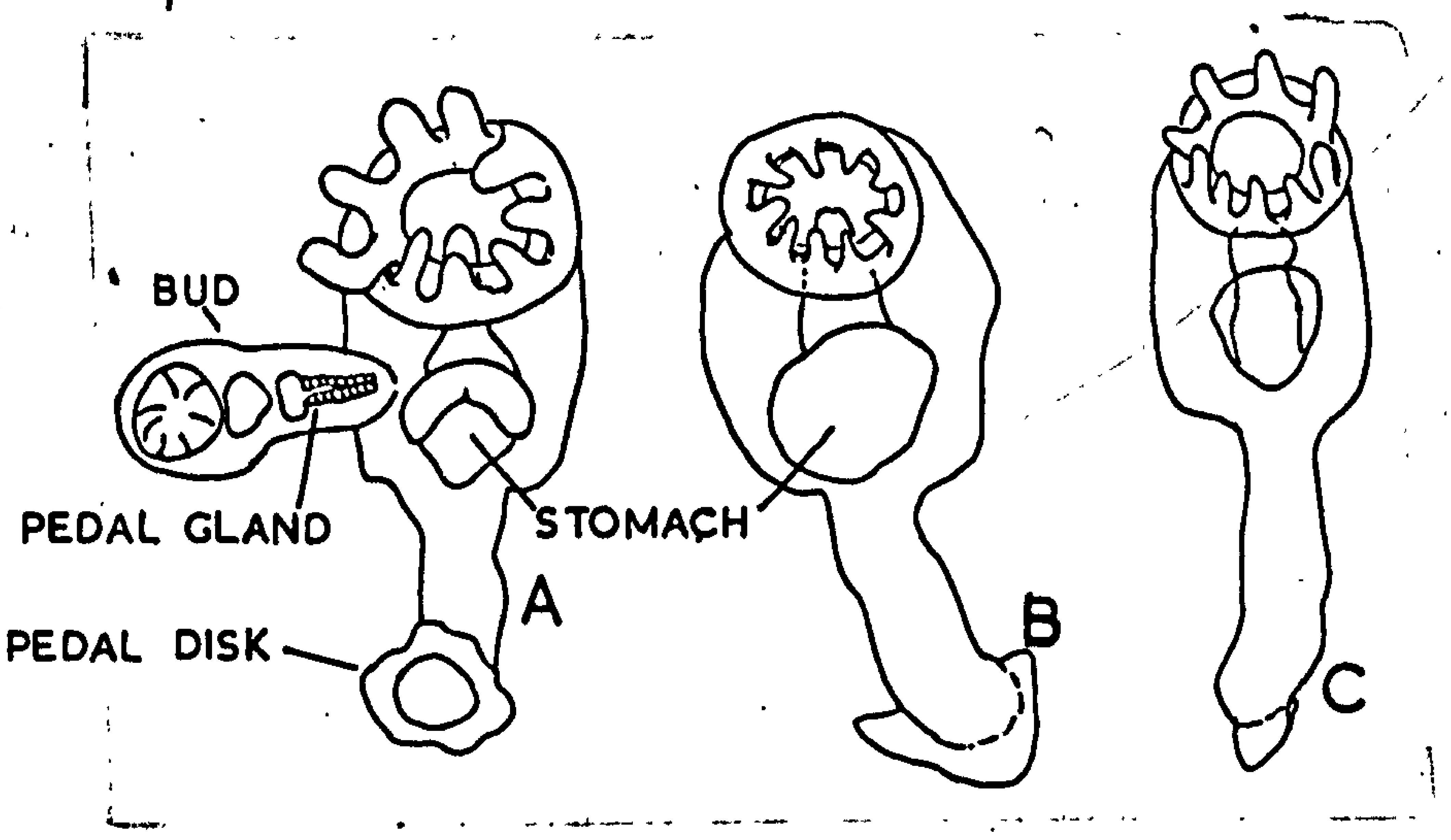

Fig. I.12. Loxosomella aryyae. A. Frontal view of specimen (Length 308 μ) with bud. B. Frontal view of contracted specimen (Length 367 μ) C. Frontal view of specimen (Length 343 μ)

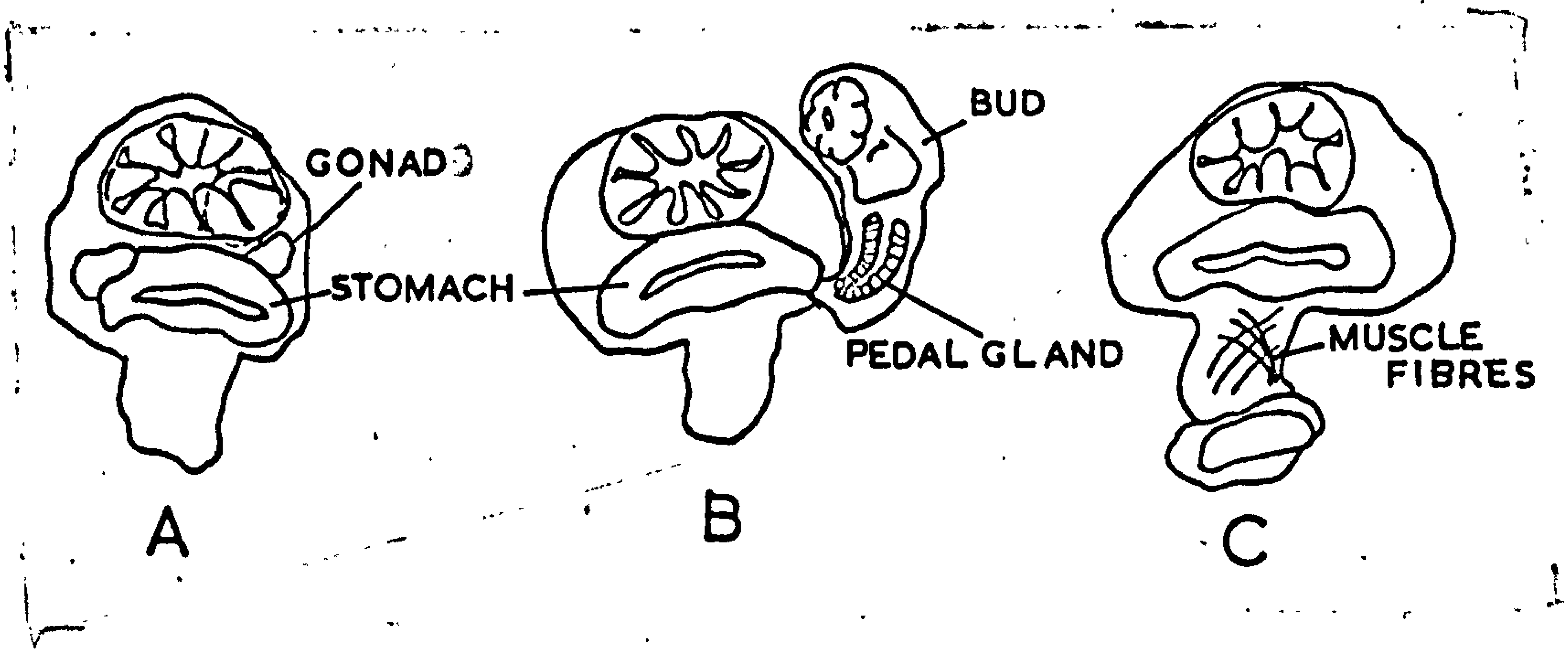

Fig I. 13. Loxosomella fungiformis. A. Frontal view of specimen (Length 158 μ) with visible gonads. B. Frontal view of specimen (Length 194 μ) with large bud. C. Frontal view of specimen (Length 202 μ). Specimens are not drawn to same scale.

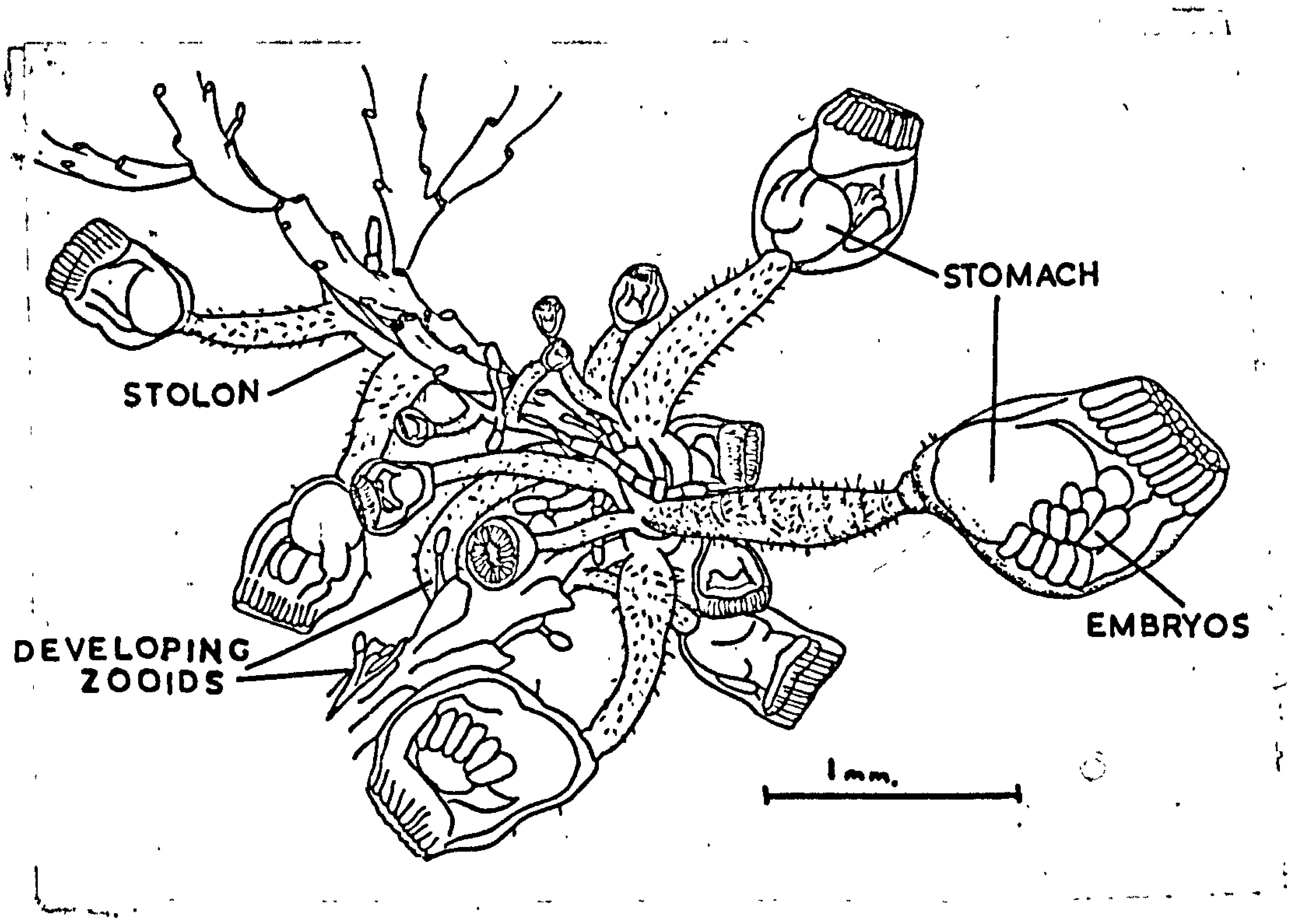

Fig I. 14. Pedicellina cernua. Part of a colony growing on Crisia eburnea. Some of the zooids contain embryos.

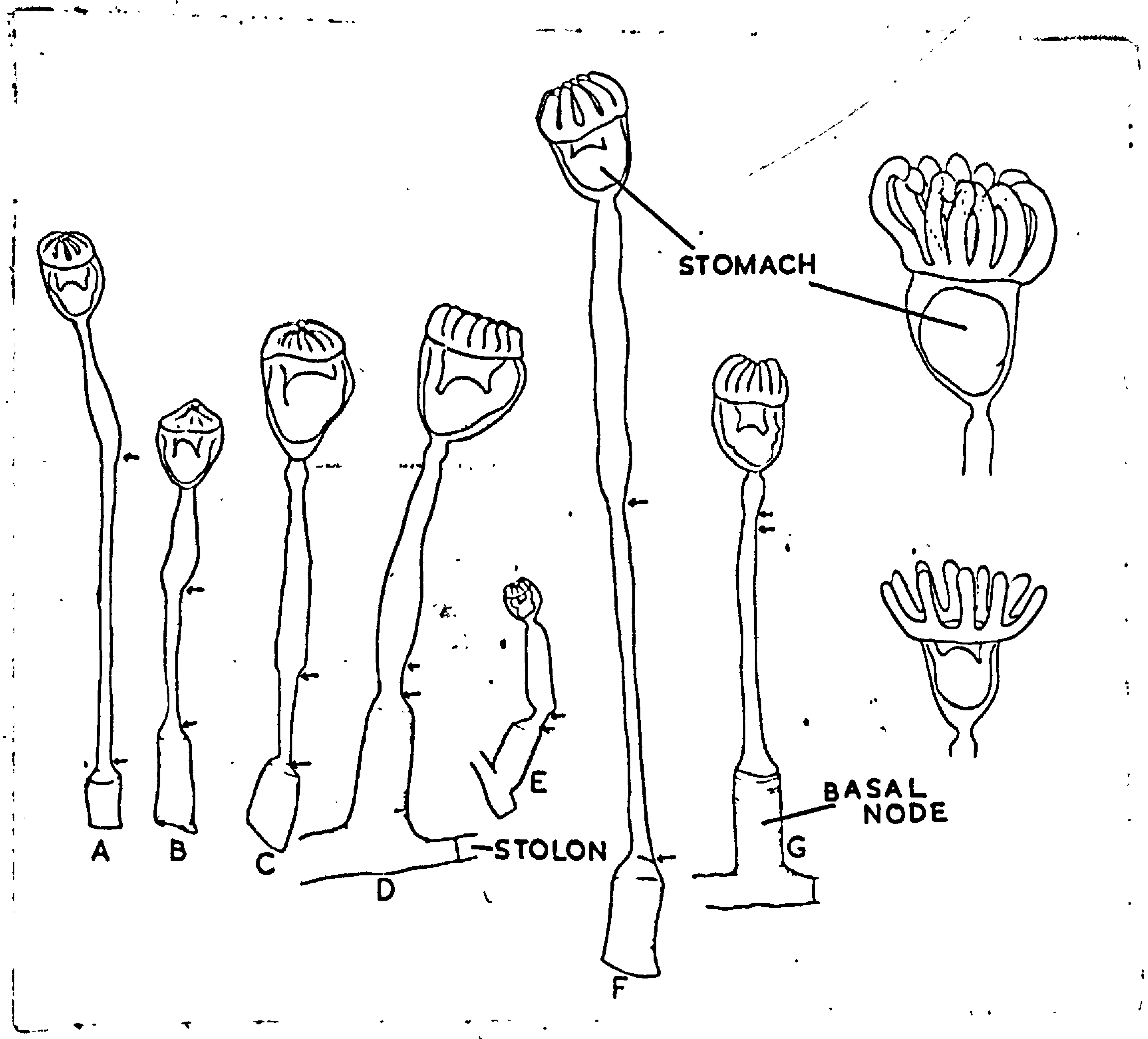

Fig I. 15. Barentsia sp. nov? Specimens A-G are all in lateral view. Of the two calyces on the right the upper is in frontal and the lower in lateral view. In specimens A-G the arrows mark the ends of the rigid part of the stalk. Specimen A measures 1188μ in total length, B 836μ, C 924μ, D 950μ, E 414μ, F 1408μ and G 1232μ.

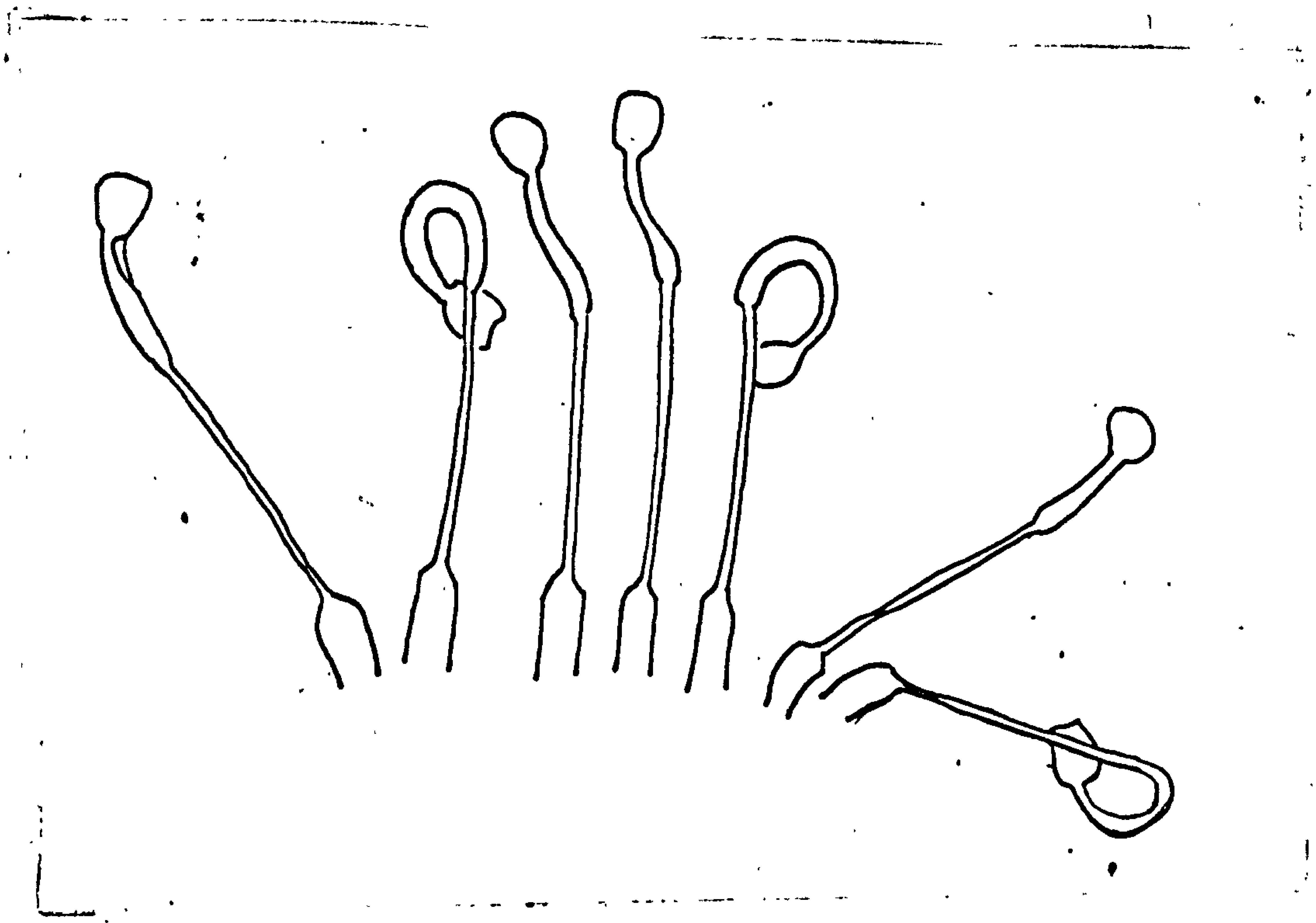

Fig I. 16. Illustrates the mobility of th stalk of Barentsia sp. nov?

PART 2

Phylum Ectoprocta

SECTION 1 : INTRODUCTION

General

The ectoprocts are a very interesting and, in some localities, an extremely abundant group of animals but probably because, (a), they are of little direct economic importance (except as ship-foulers) and, (b), they have a reputation as a difficult group, they have been neglected by many zoologists. Their reputation as a difficult group is probably a result largely of the inadequate accounts given in the majority of non-specialist text-books, and of the incomplete species lists, poor descriptions and deficient accounts given in the books by which the non-specialist is introduced to the problem of identification of the Ectoprocta. There is no good, recent, monograph on the British Ectoprocta. To use Hincks (1880) in conjunction with the key to genera given by Harmer (1910) is probably still the best means of identifying British Ectoprocta but the taxonomy used by these authors is now out-dated. A key to Manx Ectoprocta is given in Appendix IV p . Marcus (1940) "Danmarks Fauna: Mosdyr" is very good but in Danish and "Faune de France VOL 60: Bryozaires Pt.I., Entoproctes and Ctenostomes." (Bobin and Prenant 1956) is of great value to the student of ectoprocts but the figures are rather poor.

The study of the Ectoprocta, probably as a direct result of the paucity of workers on the group, has evolved slowly. Much of the work being done today is of a taxonomic nature. Johnston, Hincks, Waters, Busk, Harmer etc. laid the foundations for the study of British Ectoprocta. They accumulated large collections of species from many localities but did very little systematic collecting nor did they pay much attention to the internal structure and the biology of the various species. The result was that although the geographical range and distribution of a large number of species was fairly well known only a very little was known about the structure of the ectoproct polyp and the biology of the majority of the species had not been investigated. There was very little systematic collecting to try to establish which factors were important in controlling the distribution of ectoprocts, nor was much, apart from isolated records scattered in the literature, known of the duration, intensity or timing of reproduction in the Ectoprocta. Calvet (1900), Marcus (1921) and Borg (1923) described the anatomy of several ectoproct species and Marcus (1926) assembled the known data on the season of reproduction of European species into a single table. Within the last 20 years interest in the Ectoprocta has increased but even today most of the work on Ectoprocta is of a taxonomic nature. However attention has been focused on the biology of ectoprocts by Silen (1955 a, b) and other authors. Gautier has recently (1962) published an excellent paper on the distribution and general ecology of the Ectoprocta of the Mediterranean which is the first study to investigate the influence of various environmental factors in controlling the distribution of marine Ectoprocta. In Britain work has been largely of a taxonomic nature but Ryland (1959, 1960) studied the behaviour of ectoproct larvae; this work is being continued by Williams (Crisp and Williams 1960).

Previous work on Manx Ectoprocta.

The ectoproct section of the 1937 edition of the "Marine Fauna of the Isle of Man" is based largely on the records of Edward Forbes, T. Hincks and the L.M.B.C. workers: Prof. W. A. Herdman, Miss L. R. Thornely and J. Lomas. All these workers collected and identified Manx ectoprocts. The 2nd edition of the I.o.M.M.F. (In prep.) also includes the more recent records of identifications by N. S. Jones and M. C. Miller. A few other biologists have records in the list but, apart from some systematic dredging and shore collecting by the L.M.B.C. near Port Erin (Herdman 1900) and, indirectly, the work of M. C. Miller (1959) there has been little work recently on the Ectoprocta of the Isle of Man. The records of ectoprocts given by Jones (1940, 1951) are incomplete.

No freshwater ectoprocta have been recorded from the Isle of Man.

Aims of the present investigation

At the outset of the investigation it was realized that either of two courses could be followed, either (a) a detailed study of a small number of species could be made or (b) the ectoproct population could be studied as a whole but in a less detailed manner. It was considered that as there had been little or no systematic dredging over a period of time in any locality, and little was known of the factors controlling the distribution of ectoproct species or of the season and extent of reproduction in ectoprocts, course (b) would probably produce results of greater value. It was therefore decided to concentrate on three principal aims:

1. To investigate by systematic collecting the distribution of ectoprocts around the south of the Isle of Man and to attempt to evaluate the importance of factors such as water movement, bottom substrate (sand, muddy sand, etc.), the availability of supports suitable for ectoproct colonization (shells, hydroids etc.) depth, water movement, etc. in controlling the distribution of ectoprocts.

2. To gather information on the time and extent of the reproduction period in as many species as possible.

3. To note any general information and data on the ecology of Manx Ectoprocta.

It was felt that if these aims were satisfactorily fulfilled the results would be of value to future workers in suggesting lines of research and would provide a great deal of basic data on distribution and reproduction of the Ectoprocta.

It was hoped to investigate by experiment the settlement behaviour of ectoproct larvae (see Ryland 1959) but because of the difficulty of obtaining sufficient quantities of larvae and the shortage of time available this line of research was abandoned.

Area investigated

The area sampled is bounded by lines to the west of Niarbyl for 11 miles (17.7 km.) then south for 16 miles (25.7 km.) then east for 13 miles (20.9 km.) then north to meet the shore-line slightly to the west of Scarlett Point. This area is approximately the same as that investigated by Jones (1951) in his study of the bottom communities off the south of the Isle of Man. It encloses a wide range of environmental conditions: depth ranges from sea-level to more than 50 fathoms (90 m.), bottom substrates vary from rocky shore to offshore glutinous mud and tidal currents may be very strong or almost negligible. Map 1 shows the area sampled and indicates the distribution of bottom sediments. Many dredge hauls were taken at stations A, B, C, D and E and several from the Modiolus bed, the "Chasms" station and behind Bradda Hd., in other localities sampled during the survey only single hauls were taken. At Station A the bottom sediment is intermediate between the inshore muddy sand of Station D and the clean sand and shell of Station B. Station C and the "Chasms" station are on coarse grounds consisting of gravel and abundant shell and stones; at both tidal currents are strong. Station E is on muddy-sand in an area of little tidal movement.

Methods used in this investigation

(a) Collection of material

A wide variety of sampling mechanisms were used to obtain material.

Shore collections were made fortnightly at the period of spring tides. A number of shores were visited but most of the samples were collected at Port Erin Breakwater and particularly from the boulder beach of the outer surface of the "T-block". (The "T-block" is the local name for the inner breakwater built at the shoreward end of the main (ruined) breakwater and which runs parallel to the shore for about 25 yds.) The stones of the "T-block" beach bear rich growths of ectoprocts, sponges and ascidians. Collections of algae and stones of manageable size were made in the Ascophyllum nodosum, Fucus serratus and Laminaria zones. Observations indicated that there were no appreciable differences in numbers and species of ectoproct between the small stones taken back to the laboratory and large boulders examined on the shore.

Sub-littoral collections were obtained by a variety of means. A few collections from the shallow sub-littoral (L.W.S.T. to 100 ft. (30 m.)) were obtained by snorkel or aqua-lung diving.

Offshore samples were obtained from the research vessel "William Herdman" and the motor boat "Cypris". The gear used depended on the nature of the bottom and the species of ectoproct it was hoped to sample. The great majority of samples were obtained by using a scallop dredge lined with shrimp-netting so as to retain small material. The dredge used from the "William Herdman" measured 4' 6" across the mouth, that from "Cypris" 3' 0". Samples from "William Herdman" and "Cypris" were too similar to justify separating the results from the two boats. Usually the sample of material present in the dredge was too large to be examined *in toto*, and if this was the case the procedure varied with the function of the sample. If the sample was for work on distribution a random sample was taken after crabs, fish, echinoderms and other unwanted material had been removed. A sample usually filled a large sweet jar (= 2-3 litres volume), this was found to be about the optimum size for the purposes of the investigation. If the haul was in an area where ectoprocts were not abundant a larger sample was taken. If the haul was to obtain material for work on reproduction seasons then a random sample was not taken; shells and other supports with rich growths of ectoprocts were selected. Such samples were often larger than the samples used for distribution purposes so that more colonies of less common species would be obtained.

Some samples were obtained with an otter trawl. This instrument was particularly useful on offshore muddy grounds where the scallop dredge is inefficient. The trawl was used from the "William Herdman" and all hauls were of 1 hours duration. *Nephrops norvegicus* L. was the principal trawled support suitable for ectoproct growth but occasionally crabs and dead shell bearing ectoprocts were obtained.

The runner or ski-dredge was used twice in deep water (65 f., 120 m.). A few *Calocaris macandreae* Bell were the only supports suitable for ectoproct growth present in the ski-dredged samples.

A naturalist's dredge with a canvas bag was employed after the manner of a bucket-dredge to obtain bottom samples from some localities.

When possible weekly samples of plankton from about 6 fathom (11 m.) below the surface were obtained by using a compound plankton net from "Cypris". These samples were scrutinized for cyphonautes and other ectoproct larvae.

The positions of early "William Herdman" hauls and all "Cypris" hauls were fixed by compass bearings or landmarks and afterwards plotted on Admiralty or Ordnance Survey Charts. The installation of a Decca Navigator on the "William Herdman" in 1961 greatly facilitated the problem of fixing positions and enabled duplicate hauls to be taken from a locality without difficulty.

The depths of all "William Herdman" samples were read on an echo-sounder; depths of "Cypris" samples were estimated from Admirality charts, local knowledge or by visiting the area from which the sample was obtained in the "William Herdman" and establishing the depth by echo-sounder.

All samples were taken to the laboratory alive and in sea-water.

(b) Examination of material

The great majority of samples were examined fresh within 3 days of collection but some "distribution" samples were preserved in alcohol for a short time before examination. All samples were examined under the low-power of a binocular microscope. The data recorded depended on the function of the sample.

Samples obtained for investigations on distribution. These were examined and a species list produced for each sample; The number of colonies of each species, the number of colonies of each species on each type of support, the total Ectoprocta in the sample, the surface area of each type of support in the sample and the total surface area in each sample were recorded. From these data conclusions could be drawn as to the distribution of each species over the area as a whole, samples from the same area could be compared to check the efficiency of the sampling method, the abundance of a species or the total Ectoprocta on different supports could be compared, and the distribution of each species could be elucidated in terms of numbers/surface area ~~and~~ or as a proportion of the total Ectoprocta in a particular area or over the area as a whole.

(ii) Samples obtained for the investigation on reproduction. Hyman (1959) says that "available data indicate that ectoprocts have an annual breeding season that extends over 2 to 3 up to 5 or 6 months" and "the great majority (of ectoprocts) breed within the months April and May through October". Because of these and similar statements in the literature, it was thought that brief records of any reproductive activity observed in specimens from the 'distribution' samples would be sufficient to give information on the time and direction of reproduction in each species. However it was soon realized that many species reproduce during much or all of the year and that more detailed information was necessary. Accordingly a programme of regular samples to investigate quantitatively the intensity of reproduction in as many species as possible throughout the year was initiated. Samples for this programme were obtained from the shore and from areas A, B, C, D and E (See Map 1,) so as to obtain specimens of a large number of species from a variety of localities. Each colony of each species was examined and the data recorded included whether or not it was of mature size together with the numbers of any eggs, embryos, and empty ooecia or ovicells present. From these data various conclusions can be drawn: if eggs and empty ooecia are present this is an indication that a colony is beginning to reproduce, if numerous eggs and embryos are present reproduction is in full swing, if a few embryos are present together with empty ooecia the colony is ceasing reproduction, if only numbers of empty ooecia are present then the colony has ceased to reproduce. From the data obtained histograms of the percentage of colonies with eggs, % colonies with embryos, the % ooecia containing embryos and the number of eggs relative to the number of empty ooecia were plotted on a monthly basis. The number of small immature colonies present at different times gives an indication of the growth to maturity of new generations. Up to 100 specimens of each species present were examined from each dredge sample.

Larval colour is an important taxonomic character in the Ectoprocta (Silen 1945, Ryland 1958). Larval colour was assessed objectively for many species by using Munsell Colour Charts. These charts consist of an orderly arrangement of colour papers which serve as standards for the classification of colours. The standard papers represent equally spaced divisions of the three attributes of colour known in this system as hue, value and chroma. The hue notation of a colour indicates its relation to Red, Yellow, Blue and Green, the value notation indicates its lightness and the chroma indicates its saturation. In recording a colour by the Munsell system the symbol for hue is written first and is followed by a symbol written in fraction form, the numerator indicating the value and the denominator the chroma ($H \frac{V}{C}$). For example, a colour which is 5.0 Red in hue, 5 in value and 8 in chroma is written 5.0 R $^5/8$.

Munsell Colour Charts were used to establish larval and embryo colour by the following method. Several embryos or larvae were placed in sea-water on a cavity-slide. The appropriate colour charts were placed on a microscope stage and the slide placed on this, the whole being illuminated by a single 240 v. 60 watt bulb at about 12 inches distant at 45° above the horizontal. Care was taken to eliminate shadows from the microscope field. The colour of the embryos and larvae was assessed by looking at them under a low magnification against backgrounds of various hues, values and chromas. If their colour was between two standards the $H \frac{V}{C}$ was estimated as closely as possible.

SECTION II : RESULTS OBTAINED

Introduction

The results obtained in this investigation are given below. Data on the distribution and reproduction of each species in the Manx area is given together with available information on geographical distribution and season of reproduction in other localities. The taxonomical arrangement and the names used are those used in the 2nd edition of the "Marine Fauna of the Isle of Man". The names used in the 1st edition of the I.o.M.M.F., the 3rd edition of the Plymouth Marine Fauna, and in the work of Hincks (1880) and Gautier (1962) are given in brackets when they differ from the name used here. Keys to the Manx Ectoprocta are given in Appendix IV.

Results

PHYLUM ECTOPROCTA

CLASS GYMNOLAEMATA

ORDER CYLOSTOMATA

FAMILY CRISIIDAE

CRISIDIA CORNUTA (L.)

(Hincks (1880) and I.o.M.M.F.(1937) as Crisia)

Distribution (See Map 3 and Table 1 + 2)

Crisidia cornuta is typically present on vertical rock faces and overhangs at and below L.W.S.T. In such sites if there is little algal growth *C.cornuta*, together with *Crisia ramosa* Harmer, *Crisia denticulata* (Lm.) and *Scrupocellaria reptans* (L.), often forms a short thick turf. Such a turf is well-developed amongst the blocks of Port Erin Breakwater about L.W.S.T. Colonies of *C.cornuta* are sparsely distributed on *Laminaria* holdfasts, stones and red algae on most rocky shores.

Although most abundant on sub-littoral rock *C.cornuta* has been regularly taken in dredge samples. Colonies have been dredged on most types of supports (shells, stones, hydroids, etc.) but the majority of colonies from deeper water were attached to hydroids. *C.cornuta* has been dredged down to 30 f.(59 m) but this species is taken most frequently in shallow water (less than 20 f.(37 m)) and is most abundant where there is ample water movement.

Colonies of *C.cornuta* and other ectoprocts more usually found in shallower water were observed on the carapace of a *Maia squinado* (Herbst) trawled in 41 f.(75 m) but the crab may have migrated from shallow to deeper water after the ectoprocts had attached themselves.

Reproduction (See Fig. II.1)

Colonies of *Crisidia cornuta* bearing ovicells have been collected in all months except August, September, November and December. Ovicells are most abundant in April, May and June. Embryos have been observed from April to July with the maximum in May and June. A single colony with embryos was collected in October 1961 but no other colony displaying any reproductive activity was observed between August and December. In 1962 the first embryos were noted on April 5th and by the end of April and during May many colonies bore ovicells filled with embryos.

Ovicells containing embryos are yellow in colour but the small larvae when viewed singly are almost colourless.

Harmer (1891) found ovicells to be commonest at Plymouth during April and May. Roper (1913) found ovicells on Northumberland specimens in November.

Geographical Distribution

Arctic Ocean, Faroes, Iceland, European coast south to N.Spain; Not recorded from W.Atlantic; Pacific from Vancouver Island and Queen Charlotte Island (Marcus 1940).

FILICRISIA GENICULATA (Milne-Edwards)

(Hincks (1880) as Crisidia cornuta var geniculata)

Distribution

Previously unrecorded from Manx waters, Filicrisia geniculata has been collected twice during the present investigation. One colony, growing on Plumaria elegans Scm., was found (30.10.59) at L.W.S. near Port Erin Swimming Baths, the other, a fine colony, was attached at the base of a clump of Lomentaria articulata Lyngb collected at E.L.W.S.T. on the "T-block" of Port Erin Breakwater (10.9.60).

Reproduction

Neither of the Port Erin colonies bore ovicells.

Harmer (1891) found ovicells in Channel Island material in summer (June-August).

Geographical distribution

Arctic Ocean; Norwegian, French and Spanish Coasts, Mediterranean; S.Alaska, Vancouver Island, California; Torres St. (Marcus 1940).

CRISIA EBURNEA (L.)

Distribution (See Map 4 and Table 1+2)

Occasionally collected at L.W.S.T. under boulders and rock overhangs or among red algae, Crisia eburnea is common on sub-littoral red algae and is often present in dredge samples. More specimens have been dredged than of any other *Crisia*. Dredged colonies are usually attached to erect hydroids such as *Halecium halecinum* (L.), *Abietinaria abietina* (L.), *Hydrallmania falcata* (L.), *Sertularella* spp. etc. or to the erect ectoprocts *Flustra foliacea* (L.), *Eucratea loricata* (L.), *Cellaria* spp., etc., but colonies sometimes occur on shells and stones. *C. eburnea* has been dredged down to 37 f.(70 m) but is less common in depths greater than 25 f.(44 m). The distribution of this species may be limited by the occurrence of suitable supports.

Reproduction (See Fig. II. 2.)

Ovicells observed from March to November, embryos and larvae from April to June. The highest percentage of colonies with embryos noted was 50% in June 1961.

Harmer (1891) recorded ovicells at Plymouth from February to May with the peak numbers in March, April and May. Roper (1913) found ovicells in May, June, August and October but she sampled mainly in the summer months.

Geographical distribution

Arctic Regions, Iceland, Faroes, European Coast to Madiera; Mediterranean, W. Atlantic Coast south to Chesapeake Bay (Marcus 1940).

CRISIA ACULEATA Hassall

(Hincks (1880) as *Crisia eburnea* var. *aculeata*.)

Distribution (See Map 5 and Table 1.2)

Not previously recorded from Manx waters, *Crisia aculeata* is widely distributed in small numbers and occurs on a wide variety of supports including shells, zoophytes, stones, etc., but never forms more than a small fraction of the total ectoprocta in a sample.

Like Crisia ramosa, Crisia denticulata and Crisidia cornuta it is most common where there is considerable water movement but it extends into deeper water and areas of soft bottom substrates more frequently than the other Crisiidae. It has been collected at L.W.S.T. and was present in the deepest dredged sample (37 f., 67 m) and on the carapace of a Maia squinado trawled in 41 f.(75 m).

Reproduction

Ovicells have been observed from May-July, from October-January and in March, and embryos in January, May, July, October and December, but samples are too small to allow estimates of the period of peak reproductive activity to be made.

Harmer (1891) noted ovicells at Plymouth in April and May and in Roscoff specimens in June.

Geographical Distribution

Faroes; Finmark; Shetlands, Irish, English, Danish and French Coasts; Morocco; Mediterranean (Marcus 1940).

CRISIA DENTICULATA Lamark.

Distribution (See Map 3 and Table 1.2)

Crisia denticulata has a very restricted distribution. It is commonest in an area bounded by lines N. of Calf Stack and W. of Port Erin (This is the area in which all of the Manx Crisiidae are most abundant). It always occurs in small numbers and never forms more than 12 of the total ectoprocts in a sample. A few colonies have been collected at E.L.W.S.T. C.denticulata grows on a variety of substrates including dead shell; stones and erect zoophytes.

Reproduction

Small numbers of colonies have been examined in all months of the year but no ovicells have been seen.

Harmer (1891) recorded ovicells in Guernsey material from June to August. Gautier (1958) found ovicells in August in Sicily.

Geographical Distribution

Arctic Ocean, Iceland, European Coast to Madiera; Mediterranean; E.Coast of America to Florida and the Gulf of Mexico.

CRISIA RAMOSA Harmer

Distribution (See Map 5 and Table 1.2)

Crisia ramosa has been dredged from depths between 10 and 35 f. (18-65 m). It has not been found on the shore. It grows on a variety of supports including dead shell, stones and erect zoophytes. It is most common in areas where there is abundant clean shell and water movement but it does extend, in smaller numbers, into deeper water.

Reproduction (See Fig. II.3)

Ovicells noted in all months except February and April; embryos seen in May and from August to January. September to October is the period of greatest reproductive activity. At Plymouth Harmer (1891) found ovicells from April to August with a peak in May and June while Todd (P.M.F.) found 'breeding' colonies in February.

Geographical Distribution

Skagerrak, English, French and Spanish Coats; Azores; Cape Verde Islands; Mediterranean and Red Sea; Japan (Marcus 1940), North Carolina, Puerto Rico, Brazil (Maturo 1957).

FAMILY ONCOUSECIIDAE

Because members of this family show great variation in form and habit and cannot be certainly identified unless ovicells are present few conclusions can be drawn about their distribution in Manx waters.

PROBOSCINA INCRASSATA (Milne-Edwards)

(Hincks (1880), as Stomatopora.)

Distribution

Several specimens lacking ovicells but displaying the typical colony shape of this species were collected which almost certainly are this species. P.incrussata colonies encrust shells and stones and have a characteristic appearance. The branches divide and fuse among themselves frequently and give the compact colony a retiform appearance. The spaces between the branches are usually long, narrow and pointed at both ends and the branches frequently give rise to short cylindrical erect processes with a cellular apex. Such colonies were dredged, on shell, in the following localities: 2.5 mi(2.8 km) N of Chicken Rock (23 f; 42 m), and 7.1 mi (11.4 km) at 511°E of the Chicken Rock (37 f; 67 m).

Geographical Distribution

Arctic Region; European Coasts from Faroes to Madiera; Morocco, Mediterranean; Cape Verde Is., Azores; Queen Charlottes Land and Vancouver; Japan; and less certainly from Gulf of St.Lawrence, Tristan da Cunha, Magellan Str., New Zealand and Australia (Marcus 1940).

STOMATOPORA GRANULATA (Milne-Edwards)

Distribution

Previously recorded from Manx waters (I.o.M.M.F. 1937). Several small cyclostones lacking ovicells but which may be of this species were collected during the present survey.

Geographical Distribution

N.Atlantic E.Coast from Faeroes to Madeira; Cape Verde Is. and Mediterranean; Queen Charlottes Land and Japan (Marcus 1940).

STOMATOPORA INCURVATA (Hincks)

Distribution

Not collected in this survey; previously recorded (I.o.M.M.F. 1937) from 16-20 f.(29-37 m) E. of Calf Sound.

Geographical Distribution

?

ONCOUSOECIA DILATANS (Johnst)

(Hincks (1880) as Stomatopora; P.M.F. (1937) as Tubulipora)

Distribution

Ovicelled specimens definitely this species were dredged at the following localities: 8 mi (12.9 km) 20°S.of E. of Langness Lighthouse (22 f; 40 m), 4.5 mi (7.2 km) W 33°S of Chicken Rock (23 f; 42 m) and 1.3 mi (2.1 km) at W 31°S of Port Erin Breakwater Bouy (16 f; 29.25 m). Many other small encrusting cyclostomes which may have been this species were collected but as they were without ovicells their identity could not be confirmed.

Reproduction

Ovicelled colonies were collected in March, May and June. No embryos were seen.

Geographical Distribution

Arctic Ocean; Newfoundland Banks; Norwegian, Shetland and other European Coasts to the Mediterranean (Marcus 1940).

FAMILY TUBULIPORIDAE

A great many colonies of the Tubulipora spp. collected lacked ovicells and could not be definitely assigned to a particular species.

TUBULIPORA LILIACEA Pallas.

(Hincks (1880) and I.o.M.M.F. (1937) as Idmonea serpens)

Distribution

Specimens positively identified as Tubulipora liliacea have been collected over much of the area between 10 and 30 f (18-55 m) particularly where there is abundant hydroid growth. T.liliacea grows almost exclusively on hydroids e.g. Halecium halecinum (L.), Sertularella polyzonias (L.), Diphasia pinnaster (Ellis and Solander), Sertularia spp. and particularly on Hydrallmania falcata (L.). Up to 30 colonies have been seen on a single H.falcata colony. A few colonies were growing on shell.

Reproduction

Ovicelled specimens were observed in all months except February, May, July and August.

Roper (1913) found ovicells at Cullercoats in June, September and October.

Geographical Distribution

Arctic; all European coasts to Madeira; Mediterranean; Morocco; Azores; Woods Hole; Magellan Str; Australia and New Zealand; Vancouver, and Galapagos Islands (Marcus 1940).

TUBULIPORA PHALANGEA Couch

(Hincks (1880) and I.o.M.M.F. (1937) as T.flabellaris)

This species can only be positively identified if ovicells are present.

Distribution (~~See Map~~)

Tubulipora phalangea is the only Tubulipora spp. positively identified in shore collections. It is quite common on the undersides of rocks and stones from E.L.W.S.T. at Port Erin Breakwater and in the shallow sub-littoral. The majority of positively identified, dredged Tubulipora colonies are of this species. It has been recorded from areas A, B, C and D, from the Modiolus bed and from several other, scattered localities.

Reproduction (Fig. II 4)

Ovicells have been observed in all months and embryos from June to November and in March and April. Embryos are colourless and the ovicell must be opened to see if they are present.

Geographical Distribution

S.W. Norway and Shetlands to the Azores, Morocco and Mediterranean.

TUBULIPORA PENICILLATA (Fabr.)

(Hincks (1880) as *Stomatopora fungia*)

Not previously recorded from the Isle of Man, this species is not uncommon in several localities off Port Erin. Young specimens without erect shoots cannot be identifidd with certainty but the erect mushroom-like shoots of mature colonies are typical of the species. *T.penicillata* is distinguished from *Entalophora clavata* Busk by the following characters: the erect boletiform shoots of *T.penicillata* arise at the end of encrusting branches and usually have zooecia opening only at the top of the shoot and not at the sides while the frequently bi- or trifid shoots of *E.clavata* arise from small incrusting bases and have zooecial openings on all sides of the shoot. The ovicells are terminal in *T.penicillata* and lateral in *E.clavata*.

Distribution (~~See Map~~)

T.penicillata has been dredged down to 37 f. (68 m) but is most common in water shallower than 25 f. (46 m). It has been dredged frequently in areas A, B and C (i.e. coarser grounds) but rarely in E and D (muddy sand grounds). In areas A, B and C respectively .27, .27 and .49 colonies have been collected per 1000 cm^2 surface area of shell and stone. Table 3 shows that in areas A, B and C, *T.penicillata* is randomly distributed on the various supports available. It is, however, more common on the outer than on the inner surface of dead shell. In area A it has been frequently noted growing among the tubes of *Pomatoceros triqueter* (L.) which are very common on the outer surfaces of dead Chlamys opercularis (L.) and Modiolus modiolus (L.) shells in that area.

Reproduction

Ovicells were present in all months. Over 100 small, colourless embryos were present in the only ovicell opened (September 1961).

Geographical Distribution

Arctic Region including Labrador Coast and Gulf of St.Lawrence; Norwegian and British Coasts.

TUBULIPORA APERTA (Harmer)

Distribution

Not previously recorded from the Isle of Man. After examination of the ovicell and ooeciostome three small Tubulipora colonies growing on dead Glycymeris glycymeris (L.) shells dredged 8 mi(12.8 km) at E 20°S of Langness Lighthouse (depth 23 f; 42 m) were identified as this species.

Reproduction

Ovicells present in June.

Geographical Distribution

Arctic Ocean, Norwegian W.Coast, Faroes; Madeira; Magellan Str.; Juan Fernandez Islands (Marcus 1940).

TUBULIPORA LOBULATA Hassall

Colonies of this species can be identified only if ovicells are present.

Distribution (See Map

Specimens definitely Tubulipora lobulata have been collected at several localities including areas A and C. On the Modiolus bed several ovicelled specimens were dredged and Tubulipora colonies without ovicells but probably T.lobulata were quite numerous (about

5 per 1000 cm^2 area of shell or stone). T.lobulata has been collected only on shells and stones.

Reproduction

Ovicells have been noted in January, May and June and from August to November. Embryos have been observed in June and November.

Geographical Distribution

Faroes, Norwegian, Danish and British Coasts; Gulf of St.Lawrence and Maine coast (Marcus 1940).

FAMILY DIASTOPORIDAE

DIAPEROECIA MAJOR (Johnston)

Distribution (Map 7.)

D.major is widely distributed in depths between 20 and 35 f. (36-64 m). The numbers present are usually small but in some localities, e.g. 4.6 mi (7.4 km) N 47°W Chicken Rock and 1.3 mi (2.1 km) N 65°W of Calf Sound, D.major numbers over 4% of the total ectoproct colonies present. Numbers obtained are too small for comparisons to be made of number of specimens per unit area, but D.major forms a higher proportion of the total ectoprocts present in muddy sand areas than elsewhere (See Table). This is probably because as bottom substrate becomes muddier both numbers of ectoproct colonies and numbers of species decrease, and therefore colonies of species able to tolerate the muddier conditions make up a higher proportion of the total ectoproct population.

D.major occurs on a variety of clean hard supports (See Table 4) but is more common on smooth inner shell surfaces than on the rougher, outer surfaces; it has not been collected on algae or hydroids.

Reproduction

Ovicells were observed in June and from November to April. Developing ovicells were seen in November 1961 and embryos in February, March and June.

The embryos are colourless.

Geographical Distribution.

Arctic Ocean; European Coasts; Mediterranean; Morocco; Cape Verde Islands; Azores; Queen Charlottes Land; Galapagos Islands; Sandwich Islands; New Zealand. (Marcus 1940).

DIAPEROECIA JOHNSTONI (Heller)

(Hincks (1880), I.o.M.M.F. (1937) and P.M.F. (1957) as Stomatopora)

Very small specimens of this species and of Proboscina incrassata are very similar; a few small P.incrassata may have been identified as D.johnstoni.

Distribution

Diaperoecia johnstoni occurs with D.major in many samples, but has a slightly wider distribution than D.major in Manx waters; neither species is common. Like D.major D.johnstoni occurs on a variety of shells and stones and is more frequently present on the inner than the outer surface of dead shells. D.major zooecia are much larger than those of D.johnstoni.

Reproduction

Ovicells noted in February and March; no embryos seen.

Geographical Distribution

Plymouth (P.M.F. 1957); Adriatic (Hincks 1880)

DIPLOSOLEN OBELIA (Johnston)

Distribution (See Map 8 and Table 5)

Almost all the Diplosolen obelia colonies obtained were dredged in the area designated "Ground A – gravel and shell" by Jones (1951), very few specimens were obtained from softer grounds. It is most common on dead and living Modiolus modiolus from the Modiolus bed where it reaches a density of about 1 colony per 100 $cm.^2$ surface area and forms about 7% of the total ectoproct population. D. obelia has been collected from depths between E.L.W.S.T. and 37 f. (68 m.). It has been noted on most types of dead shell.

Reproduction

Ovicells were observed in November and April, embryos in April.

Georgraphical Distribution

Arctic Region; European coasts south to Morocco; Mediterranean; Maine Coast; Cuba; Japan; Sandwich Island; Vancouver.

DIASTOPORA PATINA (Lamark)

Distribution (See Map 9 and Table 5)

Diastopora patina is widely distributed. It has been collected on stones at L.W.S.T. and dredged on a wide variety of supports. About 10% of D. patina colonies grow on erect zoophytes particularly the hydroids Hydrallmania falcata and Sertularia spp, and the ectoprocts Eucratea loricata (L.), Cellaria spp, Scrupocellaria spp. etc. More colonies of D. patina grow on the inner than on the outer surface of dead shells. D. patina usually makes up only a small proportion of the total ectoprocta in a sample but in some samples it is more common. 16.5% of the ectoprocts present in a sample from 3.6 mi (5.75 km) N. of Thousla Beacon were of this species. The highest density recorded for this species is 15.3 colonies per 1000 $cm.^2$ hard surface area is one sample from area C. The mean densities for areas A (10 samples), B (15 samples) and C (10 samples) are .6, 1.5 and 2.3 colonies per 1000 $cm.^2$ hard surface respectively.

D. patina has been noted on the carapace of specimens of Macropodia longirostis (Fabr.) and Hyas coarctatus Leach.

Reproduction

Ovicells have been noted in January, March, June-August and November. Embryos were seen in June, July and August.

Geographical Distribution

Kara Sea; N. Coast of Norway; S. Coast of Labrador; Newfoundland Banks; Faroes and all temperate European Coasts to the Azores; Mediterranean; Morocco; Tristan da Cunha and Atlantic Patagonian Coast; Queen Charlottes Land; Vancouver Island; a variety from Japan (Marcus 1940).

DIASTOPORA SUBORBICULARIS (Hincks)

Distribution (See Map No. 11 and Table 6)

Diastopora suborbicularis is widely distributed in the area sampled. It is most abundant on grounds where there is abundant shell and gravel, most of there grounds are shallower than 25 f. (45.7 m.) but where they extend deeper than this e.g. south and south west of the Calf, the distribution of D. suborbicularis parallels their extension. D. suborbicularis has been collected from L.W.S.T. and was present in the deepest dredged sample (37½ f.; 68.5 m.). It is one of the commoner ectoprocts in the Manx area, in many inshore samples it constitutes between 5 and 10% of the total ectoprocts present.

It does not grow on algae or zoophyts but is more or less randomly distributed among various types of shells and stones. It is sensitive to surface texture and on shells where the outer surface is rough e.g. Chlamys opercularis; it is most common on the inner smoother surface but on shells such as Ensis spp or Lutraria lutraria (L), where both surfaces are smooth it occurs in approximately equal numbers on both surfaces. (Table 6).

Reproduction

Ovicells are present throughout the year. Larvae and embryos have been noted in September, October, and from December to April. Early in the development of embryos the ovicell contents are yellowish but later becomes colourless.

Geographical Distribution.

Arctic Ocean; Faeroes; European Coast to Morocco; Mediterranean; Queen Charlottes Land? (Marcus 1940).

FAMILY ENTALOPHORIDAE

ENTALOPHORA CLAVATA (Busk)

Distribution

Previously unrecorded from the Isle of Man, five specimens were collected in this investigation. Four were dredged at Area C and one about ½ mile (.8 km.) W of Bradda Hd. All were growing on dead shell.

Reproduction

No ovicells were present.

Geographical Distribution

Arctic Ocean; European Coasts to Madeira; Mediterranean; Canary Island; Vancouver (Marcus 1940).

FAMILY LICHENOPORIDAE

LICHENOPORA RADIATA (Audouin)

Distribution

Not collected in this survey, Lichenopora radiata is recorded in the I.o.M.M.F. (1937) from the mouth of Port Erin Bay.

Geographical Distribution

British Coasts; Madiera; Mediterranean; Samoa; Australia; Japan; California.

LICHENOPORA HISPIDA (Fleming)

Distribution (See Map 12-13 and Table 7-8)

Present in almost every sample Lichenopora hispida has been collected at depths from L.W.S.T. to 42½ f. (78m.). It is a common species and over much of the area more than 5 colonies per 1000 cm.^{2} surface area of support are present and in many samples between 5% and 10% of the total ectoprocts present are of this species. The maximum density recorded for this species was 41 colonies per 1000 cm.^{2} surface area in a sample from 7.6 mi. (11.2 km.) at S 22°E. of the Chicken Rock where over 25% of the ectoprocts were L. hispida. About 1% of L. hispida colonies grow on hydroids, the remainder on stones and various shells, L. hispida is more common on the inner than the outer surface of dead shells, this is particularly true of shells in which the outer surface is much rougher than the inner (Table 7).

Reproduction

Large colonies becoming ovicells have been observed in all months. Embryos and larvae were noted in February, March, April, June and September. Ancestrulae and tiny colonies were seen in April and May.

Geographical Distribution

Arctic Ocean; Faroes, Norway and W. European Coasts to Madeira; Mediterranean; W. Coast of Labrador, Maine Coast; Florida; Queen Charlottes Land; Vancouver; Tristan da Cunha; Australia and New Zealand.

LICHENOPORA VERRUCARIA (Fabr.)

Distribution

Not collected in this survey. Recorded in the I.o.M.M.F. (1937) as present off the E. coast of the Isle of Man.

Geographical Distribution

Throughout Arctic Region; American Atlantic Coast to Woods Hole; Iceland; Faroes; Norwegian and other European Coasts; Mediterranean; Alaska; Queen Charlottes Land; Vancouver; N. Japan; Cape of Good Hope; Loyalty Islands.

ORDER CHEILO-CTENOSTOMATA

FAMILY AETEIDAE

AETEA ANGUINA (L)

Distribution (See Table 9, Map 14.)

This species is not widely distributed but ~~it is~~ in some localities ~~and~~ it is ~~a~~ of regular occurence. Its distribution appears to be correlated with the presence of suitable erect zoophyte supports. Over 70% of the colonies collected were growing on hydroids or erect ectoprocts, particularly on the hydroids Hydrallmania falcata and Sertularia spp. and the ectoprocts Eucratea loricata (L). and Cellaria spp. the remaining colonies were on shells and stones. A. anguina has been collected from depths between L.W.S.T. to 37 f. (69m.) but it is rare below 27 f. (49 m.). It has been dredged regularly in areas A, B, and C where almost every erect zoophytes bears a colony of this species.

Reproduction

Embryos were observed only once (August 1961) they are golden-yellow.

Geographical Distribution

Present in all areas except polar seas (Cantier 1962.).

AETEA SICA (Couch)

(Hincks (1880) and 1.o.M.M.F. (1937) as *Aetea recta*).

Distribution (See Map 15 and Table 9,10 and 11).

Aetea sica is widely distributed in areas of sandy shell ground with abundant dead shell but is less common elsewhere. In only a few samples were over 10 colonies per 1000 cm.^{2} surface area recorded and it rarely makes up more than 5% of the ectoprocts present in a sample but is frequently present in small numbers. *Aetea sica* occurs on a variety of supports but principally on dead shell. It is much more common on the outer than the inner surface of dead shell (Table 10). It has been collected from L.W.S.T. and dredged down to 37 f. (69 m.).

Reproduction (See Figure 5.).

Very few "ooecia" were observed without embryos, they may be either withdrawn or discarded after larval release. In 1961 embryos were present from May to November with the highest number of colonies (15%) with embryos in August. Only a few (10-20%) of the individuals in a colony bear embryos at any time.

Embryos are golden-yellow in colour.

Geographical Distribution

Present in all except Polar seas.

AETEA TRUNCATA (Landsborough)

Distribution (Map 14)

A few colonies of this species have been collected, mostly in areas A, B, and C. It is a very variable species (Hincks 1880 P.9) and both dwarf and erect "varieties" as well as the "normal" form have been seen. The erect type has been observed most often straggling over erect hydroid and ectoproct colonies, dwarf and "normal" colonies were collected on dead shells.

Reproduction

No reproductive activity was noted.

Geographical Distribution

Widely distributed in all but Polar seas.

FAMILY SCRUPARIIDAE

SCRUPARIA CHELATA (L) and Sc. AMBIGUA (d'Orbigny)

(Hincks (1880) and I.o.M.M.F.(1937) as Eucratea chelata)

Distribution (See Map 16)

Colonies of both species are small and some may have been overlooked but neither species appears to be common. On the shore tufts of Plumaria elegans Schm. usually bear colonies of one or both species. In dredge samples both species have been collected on a variety of dead shells and erect zoophytes; Sc. chelata is recorded in equal numbers from shells and zoophytes while Sc. ambigua is twice as common on zoophytes than shell but the numbers collected are very small.

Reproduction

Scruparia chelata ooecia were observed in August and September; Sc. ambigua ooecia were seen in August.

Geographical Distribution

Sc. chelata: British Coasts; Roscoff; New Zealand.

Sc. ambigua: British Coasts; Sweden; Falkland Islands; Galapagos Island. Coast of Patagonia; Australia; Tasmania; New Zealand.

EUCRATEA LORICATA (L).

(Hincks (1880), I.o.M.M.F. (1937) as Gemellaria)

Distribution (See Map 17 and Table 12)

Eucratea loricata has a limited distribution, being confined to areas of abundant dead shell and considerable water movements. It is most common at area C but also occurs in and around areas A and B. It has been collected on a wide variety of supports but occurs most often on the outer surface of dead Chlamys opercularis, Glycymeris glycymeris and Pecten maximus. Small colonies have been noted on Cellaria spp. and other zoophytes.

Reproduction.

I have found no previous published record of any reproductive activity in this species. One colony of Eucratea loricata collected in March 1962 bore membranous "ooecia" similar to those of Aetea spp. The "ooecia" are transparent, membranous and placed singly at the distal edge of the operculum, they appear to extend into the zooecial cavity. The embryos are whitish colours and in various states of development.

The difference between the endozooecial ovicells of Scruparia spp. and the "ooecia" Eucratea loricata indicates that these genera should be placed in separate families.

Geographical Distribution

Arctic Region; European Coasts to Channel; N. American Coast from Arctic to Woods Hole and Vancouver.

FAMILY MEMBRANIPORIDAE

MEMBRANIPORA MEMBRANACEA (L)

Distribution

Membranipora membranacea is abundant on all rocky shores, it forms extensive sheet-like colonies on the fronds of *Laminaria* spp. from L.W.S.T. down to the lower limits of the *Laminaria*. It also occurs but less frequently on *Fucus serratus* L. and on broad-fronded red algae such as *Gigartina*, *Chondrus* and *Delesseria* spp. A few colonies growing on stones have been collected on the shore and a colony was present on a stone dredged in 23 f. (42 m.).

The annual growth cycle of *M. Membranacea* is correlated with that of the *Laminaria* spp. At the end of summer (September - October) the majority of *Laminaria* plants bear large colonies of *M. membranacea*. During the winter growth of both organisms is checked and some of the *Laminaria* fronds, with their *M. membranacea* colonies, are torn from the stripes during the winter gales. Growth of the new *Laminaria* frond begins in January and continues into June and July. In early April "rapid" growth on the part of those *M. membranacea*, colonies remaining on the old fronds enables some of them to colonise the new growth before the remainder of the old frond are cast in April or May. The overwintering colonies of *M. membranacea* are sexually mature and release many eggs during April, May, and probably later months (not examined for eggs). The length of time spent in the plankton by the cyphonautes larvae of M. membranacea is not known. Settlement begins in late May and continues until August so that by the end of summer most Laminaria fronds bear M. membranacea colonies. This species is very quick-growing; Lutand (1957) recorded a growth of 1-2 cm. in 4 days during laboratory experiments.

Reproduction

There are no brood protection mechanisms in M. membranacea the egg is laid and develops into the cyphonautes larva known as Cyphonautes schneideri Lohm.

Zooids were scrutinized for eggs and sperm between January and June 1962. Sperm morulae were observed in February and eggs and sperm in April, May and June. Most reproducing zooids contained between 10 and 20 buff coloured eggs but over 50 were present in one zooid. When mature sperm are present they often completely pack the zooecial cavity. Some of the zooecia containing eggs also contained a few spermatozoa.

Egg-laying was observed on two occasions in May 1962, on both occasions the pattern of events was the same. The egg-laying process was watched through a binocular microscope and is as follows: the zooid extends its tentacles in the normal manner and an egg can be seen at the base of the inter-tentacular organ, the egg enters the lumen of the tube and moves slowly towards the aperture until it begins to protrude when it is quickly extruded and swept away by the tentacular currents of the zooid and its neighbours. The zooid does not usually retract while the egg is in the inter-tentacular organ but if it does retract the egg is expelled almost immediately on re-extension. The time between entering the lumen of the inter-tentacular organ and final expulsion varied between 15 seconds and one minute 40 seconds of which only a few seconds were spent in expulsion from the aperture of the inter-tentacular organ, the

the rest being spent in passing along the lumen of the tube. The eggs are laid in quite rapid succession; one zooid laid 9 eggs in less than 20 minutes and 12 zooids laid a total of 42 eggs in the same time. The eggs are distorted into a cigar-shape during laying but soon become a flattened oval shape. They measure from 80 to 120 μ in length, about 80 μ in width and about 30 μ in depth. Soon after laying a translucent membrane surrounding the egg becomes visible. Several hundred eggs were kept in aquaria for a few days but apart from a few initial cleavages little development took place.

Plankton samples were collected throughout the year by towing a compound plankton net at a depth of about 6 f. (11 m.) for 10 minutes just off Port Erin Breakwater. Cyphonautes schneideri was collected in all months from February to November and in greatest numbers in June, July and August. Newly settled larvae and small colonies were collected in largest numbers in June, July and August.

Several Cy. schneideri kept in the laboratory metamorphosed into the typical "double-ancestrula" of M. membranacea on 20 7.60, after a week all the colonies consisted of the ancestrula plus at least 4 zooids, after two weeks from 7-13 zooids were present and after 5 weeks over 50 individuals were present in all the colonies and rapid growth was continuing.

At Plymouth Atkins (1955) found C. schneideri throughout the year but most commonly in autumn. Hastings (PMF 1957) observed eggs, sperm and egg laying in Plymouth material in June.

Geographical Distribution

European Coasts; South to Madeira and Morocco; Mediterranean American Coasts from Labrador to Tortugas and from Alaska to California; S. Africa, Australia, New Zealand, Chile.

<u>ELECTRA PILOSA</u> L.

<u>Distribution</u> (See Map 18)

The species has a wider distribution than any other ectoproct in Manx waters, it has been collected from L.W.N.T. level on rocky shores down to 47 f. (81 m.) where it frequently occurs on <u>Nephrops norvegicus</u> (L). Offshore it is present almost everywhere but has not been collected from the coralline and soft grounds south and west of Niarbyl. It has been noted on a wide variety of dead shells, stones, hydroids (particularly <u>Hydrallmania falcata</u> and <u>Sertularia</u> spp), erect ectoprocts (e.g. <u>Cellaria</u> spp, <u>Eucratea loricata</u> etc.) on several species of large crustacea (<u>Nephrops norvegicus</u>, <u>Cancer pagurus</u> (L). <u>Homarus gammarus</u> (L) <u>Palinurus elephas</u> (Fabr.), <u>Macropipus</u> spp. <u>Ebalia</u> spp. and a variety of spider crabs etc.,) On the other shore <u>E. pilosa</u> grows on <u>Fucus serratus</u>, <u>Laminaria</u> spp., <u>Chondrus crispus</u> Lyngb, <u>Gigartina stellata</u> Batt., <u>Plumaria elegans</u>, and numerous other algae as well as boulders and stones.

<u>Electra pilosa</u> is frequently present on the shells inhabited by the hermit crab, <u>Pagurus bernhardus</u> L. Throughout the investigation data were collected on the occurrence of <u>E.pilosa</u> on <u>Buccinum</u> shells inhabited by <u>P. bernhardus</u> on live <u>Buccinum</u> shells, and on empty <u>Buccinum</u> shells. The data obtained are included in Table 13 and can be summarized as follows: (a) <u>E. pilosa</u> is twice as common on shells inhabited by hermits than on empty shells without hermits (although at some time hermits may have lived in the empty shells, (b) <u>E. pilosa</u> is more often present on empty shells with hermits than on empty shells with hermits than on empty broken shells unsuitable for hermits, (c) <u>E. pilosa</u> is found more frequently on the inner than the outer surface of <u>Buccinum</u> shells. Typically <u>E. pilosa</u> either lines the inner surface of the mouth of the shell or grows immediately outside the mouth; it is rarely present high on

the spire of the shell. Two colonies were observed on the opercula of live Buccinum undatum. These results indicate that a loose association may be present between the ectoproct and the hermit crab; both species can, and do, occur without the other. It may be that crab-inhabited shells are cleaner than either empty shells or living Buccinum shells and are therefore more likely to be colonized by the ectoproct, or that the water movements caused by the hermit crab may encourage settlement on the crab-inhabited shells. It is hoped that the opportunity to perform settling experiments to investigate this association will arise in the future.

Reproduction

The larva of Electra pilosa is Cyphonautes compressus Ehr: (Atkins 1955) which was present in plankton samples from October to April and in July and in greatest numbers in November, February and April. Small colonies were seen in October, November, May and July.

Atkins (1955) found this cyphonautes at Plymouth throughout 1953, but chiefly in the autumn. Marcus (1940) records it from the Baltic and North Sea particularly in the winter months and (1925) at Naples from February to April.

Geographical Distribution

Arctic; N. Atlantic south to Morocco and Chesapeake Bay; Mediterranean; Red and Arabian Seas; S. Africa; Australia and New Zealand (Marcus 1940).

PYRIPORA CATENULARIA (Fleming)

(Hincks (1880) and 1.0.M.M.F. (1937) and Marcus (1940) as Membranipora).

Distribution (See Map 19 and Tables 14 and 15)

Pyripora catenularia is widely distributed in small numbers on coarse grounds where it has been collected on a variety of dead shells (e.g. Chlamys opercularis, Modiolus modiolus, Pecten maximus, Glycymeris glycymeris etc.) and stones. It occurs frequently on small pieces of gravel and shell fragments of about 2-5 cm.^{2} surface area. One colony was observed on Hydrallmania falcata. It has been collected in various depths from 12-30 f. (22-55 m.) but is most common where there is considerable water movement e.g. around the Calf. It has not been collected from muddy grounds.

A few colonies of P. catenularia have been observed with erect shoots rising from the incrusting part of the colony. On erect shoots the zooids are longer and narrower than normal Pyripora catenularia zooids. I have not found any previous description of erect shoots in P. catenularia.

Reproduction

No reproductive activity was noted.

Geographical Distribution

Faroes; Norway; British Coasts; Biscay; Azores; Mediterranean; Labrador; Gulf of St. Lawrence; Zanzibar; S. Australia.

CONOPEUM RETICULUM (L)

(Hincks (1880 and I.o.M.N.F. (1937) as Membranipora Lacroixii (Aud.).

Distribution

Previously recorded from tide-pools at Ramsey (I.o.M.N.F. 1937) Conopeum reticulum was not collected during the present survey.

Geographical Distribution

Scottish, Danish and European Coasts south to Mediterranean; Azores; Canary Islands; Cape Verde Islands; Gulf of St. Lawrence and temperate coast; N. America Coasts; Brazil; Red Sea; Indian Ocean; Australia; New Zealand; Japan; American W. Coast from Vancouver to California.

ALDERINA IMBELLIS (Hincks)

(Hincks (1880), I.o.M.H.F. (1937) and Marcus (1940 as Membranipora).

Distribution (See Maps 20 & 21 and Table 15 and 16).

Alderina imbellis is widely distributed. It is most common on muddy sand grounds particularly those N.W. of Bradda Hd. where more than 20 colonies per 1000 cm.^{2} support surface area were noted; it is scarce on coarse grounds with much water movement. Alderina imbellis was trawled in water over 40 f. (73 m.) deep and dredged in less than 10 f. (18 m.) on the muddy grounds (Area D.) close to Port Erin Breakwater.

A. imbellis occurs on a wide variety of dead shells and stones and on live Buccinum undatum, Anomia spp., Pecten Maximus etc. It is more common on smooth than on rough surfaces. 5 times as many colonies are present on the inner than the outer surface of Chlamys opercularis shells but equal numbers occur on the two surfaces of Cyprina islandica shells. It has not been collected on zoophytes.

Reproduction (See Fig II 6+7)

Ooecia embryos and eggs have been observed in all months. From June 1961 to May 1962 over 50% of well-grown colonies bore embryos in all months except July (44%) and September (30%) and in the same period over 30% of the ooecia contained embryos in the months September to May and over 75% in February and May.

A second, well-developed egg may be present in zooid while the previous embryo is still in the ooecium.

Embryo colour varies between white and pale buff (Munsell notation 10.0 YR $\frac{8.5}{6}$).

Geographical Distribution

Norway to Biscay; Tunis; Phillipines (Marcus 1940).

ALDERINA SOLIDULA (Hincks.)

(Hincks (1880) as Membranipora).

Distribution

One colony was identified on a shell dredged 3.5 m. (5.6 km) at N. 82°E. of the Chicken Rock (depth 22 f. 40 m.).

Reproduction

No data.

Geographical Distribution

Gautier (1962) lists this species as recorded from the Mediterranean by waters but does not include the reference in his bibliography.

CALLOPORA DISCRETA (Hincks.)

(Hincks (1880) and 1.o.M.M.F. (1937) as Membranipora).

Distribution

This species was recorded by Herdman (1896) from "E. of the Calf Sound, 16-20 f." It was not collected in this survey.

Geographical Distribution

No data.

CALLOPORA LINEATA (L)

(Hincks (1880) and 1.o.M.M.F.(1937) as Membranipora)

Distribution (Map 22 & Table 15)

Callopora lineata occurs on most rocky shores below M.T.L. It forms large colonies incrusting the undersides of boulders and stones; it also grows Laminaria holdfasts and occasionally on red algae, e.g. Plumaria elegans. Callopora lineata is extremely abundant on the fronds of Laminaria saccharina where it forms circular colonies in the concavities of the frond surface; by the end of summer (September-October) several thousand colonies may be present on a single frond. C.lineata is present in small numbers on dead shell on much of the coarse ground around the south of the Isle of Man and has been dredged in 37 f.(69 m).

Reproduction (See Fig. II 8 and II 9)

Ooecia, eggs and embryos are present throughout the year; reproductive activity is at its highest in June, July and August and its lowest from October to December.

Gautier (1962) recorded ooecia in December in the Mediterranean.

Eggs, embryos and larvae are red (Munsell colour 5 OR $\frac{5}{10}$)

Geographical Distribution

Arctic Region; N.Atlantic Coasts to Madeira and Florida; Mediterranean; S.Alaska to Vancouver (Marcus 1940).

CALLOPORA DUMERILI (Aud.)

(Hincks (1880), I.o.M.M.F. (1937) and Marcus (1940) as Membranipora.

Distribution (See Map 23) ~~and Table~~

Callopora dumerili is present in small numbers on all coarse grounds where it occurs most frequently on the inner surfaces of dead shells. A few colonies were collected on stones from E.L.W.S.T. at Port Erin Breakwater and the Calf Sound. It has never been collected on algae.

Reproduction (See Fig. II 10 & II 11)

Ooecia, embryos and eggs are present throughout the year, peak reproductive activity is from June to November.

Gautier (1962) recorded ooecia in March, April, June-September and in December, embryos in May and June and ancestrulae in February in the Mediterranean.

Eggs, embryos and larvae are yellow-orange (Munsell 2.5 YR $\frac{6-7}{10}$). A large egg may be present in a zooid while the previous embryo is still in the ooecium.

Geographical Distribution

European coasts from S.W.Norway to Madeira; Mediterranean; Morocco; Gulf of St.Lawrence; Maine; Tristan da Cunha? (Marcus 1940).

CALLOPORA AURITA (Hincks)

(Hincks (1880), I.o.M.M.F. (1937) and Marcus (1940) as *Membranipora*).

Distribution

Callopora aurita has been collected only from the shore (Port Erin Breakwater, Spaldrick, Fleshwick, The Sound, Perwick, etc.) where it forms extensive colonies on the undersurface of stones and boulders. It occurs from the mid-*Ascophyllum* zone through the *Laminaria*-zone fringe, its lower limit has not been determined but it was not dredged. It does not grow on algae.

Reproduction (See Fig. II 12 and II 13)

Embryos were noted from August to February and a few in April. Peak reproductive activity is in October to January.

Embryos, eggs and larvae are white.

Geographical Distribution

European Coasts from Faroes to S.England; Azores; Hudson Str.; Maine and Cape Cod; Var *norvegicus* occurs in N.Norway and the White Sea.

CALLOPORA CRATICULA (Hincks)

(Hincks (1880), I.o.M.M.F. (1937) and Marcus (1940) as *Membranipora*.)

Distribution (See Map 23)

Callopora craticula has a limited distribution; it is confined to shallow (less than 20 f.(37 m)), inshore, muddy grounds, e.g. those close to Bay Fine, Port Erin Breakwater and Bradda Hd. A few colonies grow mixed with Callopora lineata on Laminaria saccharina fronds but most of the colonies collected were growing on the inner surface of dead shells. It is not common.

Reproduction (Figs 14 and 15)

Embryos have been noted in all months and eggs in all except June and February (months in which very few colonies were collected). Early embryos are red (Munsell 10.0R$\frac{6}{8}$) but become more orange as development proceeds (7.5R$\frac{6}{10}$ to 2.5 YR $\frac{6}{12}$).

Geographical Distribution

Throughout Arctic Ocean and in N.Atlantic south to Cornwall and Woods Hole.

AMPHIBLESTRUM FLEMINGI (Busk)

(Hincks (1880), I.o.M.M.F. (1937) and Marcus (1940) as Membranipora)

Distribution (See Map 24 and Tables 15 and 16)

Amphiblestrum flemingi occurs on coarse grounds in small numbers. It has been dredged down to 37 f.(69 m) and twice recorded on stones from E.L.W.S.T. Over a quarter of the A.flemingi colonies collected were growing on live lamellibranchs, particularly Chlamys opercularis, Modiolus modiolus and Anomia spp. In area B, living Chlamys opercularis are common and over 30% of them bear at least one colony of this ectoproct, which occurs on both upper and lower shells. A.flemingi also occurs on a great variety of dead shells and usually is most common on the inner surface, but on Chlamys opercularis it occurs more commonly on the outer surface probably as a result of the high settlement on living C.opercularis.

Reproduction (See Fig. II 16 and II 17.)

A high percentage of colonies with embryos was noted throughout the year. The % of ooecia containing embryos is highest in summer (June-November).

An egg is frequently visible in a zooid while the previous embryo is still in the ooecum.

Eggs, embryos and larvae are orange (Munsell colour 5.0-10.0 YR $^7/_{6-10}$).

Geographical Distribution

Arctic Ocean and N.Atlantic south to Biscay and Woods Hole; Mediterranean.

AMPHIBLESTRUM SOLIDUM (Packard)

(Hincks (1880), I.o.M.M.F. (1937) and Marcus (1940) as Membranipora trifolium)

Distribution

Not collected in this survey; recorded by Herdman (1893) from 8 ml.(13 km) west of Fleshwick.

Geographical Distribution

N.Norway, Faroes, Shetland, Rockall Bank, N.Scotland, Ireland; Adriatic.

AMMATOPHORA NODULOSA (Hincks)

(Hincks (1880) and I.o.M.M.F. (1937) as Membranipora.)

Distribution

Not collected in this survey, recorded by Herdman (1896) from E. of the Sound 16-20 f.(29-37 m).

Geographical Distribution

Antrim, S.Devon (Hincks, 1880).

<u>CAULORAMPHUS SPINIFERUM</u> (Johnston)

(Hincks (1880), I.o.M.N.F. (1937) and Marcus (1940) as *Membranipora*.)

Distribution

Except for one colony dredged on a dead *Chlamys* shell in area C, this species has been collected only on rocks, stones and *Laminaria* holdfasts on the shore. It extends from the mid-*Ascophyllum* zone through the *Laminaria* zone fringe; its lower limit was not determined. It is a common species and forms large neat colonies spreading over stones.

Reproduction (See Fig. II 18.)

The ooecia are very small (Bobin and Prenant, 1961). *C.spiniferum* reproduces from August to May with a well-marked peak in reproductive activity from January to March when up to 80% of the colonies contain embryos.

Embryos and eggs are orange in colour (early stages are Munsell 2.5 YR $\frac{6-7}{12-14}$ later stages 7.5-10.0 YR $\frac{7}{6}$).

Geographical Distribution

Faroes, Shetland, British and N.French coasts; W.coast of N.America from S.Alaska to California; Galapagos Is. and Chile. Records from the Arctic are of doubtful validity. (Marcus 1940)

<u>ROSSELIANA ROSSELII</u> (Aud.)

(Gautier (1962) shows that two species have been confused under *R.rosselii*; the specimens collected in this survey are of *R.rosselii* (Aud.) *sensu.stricto*.)

Distribution

Only 7 *R.rosselii* colonies were collected; one each from area B, area D, the mouth of Fleshwick Bay (18 f, 33 m) and 3.2 ml (5.1 km) S 23°W of Port Erin Breakwater Bouy, and three colonies from area A. All were on dead shell.

Reproduction

No reproductive activity was noted.

Geographical Distribution

Temperate European seas; Mediterranean; British Columbia ? (Goûtier, 1962).

FAMILY FLUSTRIDAE

FLUSTRA FOLIACEA (L.)

Distribution

I cannot agree with the I.o.M.M.F. (second edition) which states that Flustra foliacea is "common on shells and stones on coarse grounds around the S. of the Island, 12-30 fm". The only locality where I have found F.foliacea to be common is area C where as many as 20 large colonies may be obtained in a single dredge haul; elsewhere more than 1 or 2 colonies are rarely present in a dredge haul and the majority of hauls contain no F.foliacea. At area C there is abundant clean shell and stone and strong tidal flow. F.foliacea colonies grow on dead shell and stones.

Local knowledge tells of a bed of F.foliacea E. of Langness but two exploratory trips failed to locate it; this bed may, like area C, be of limited extent.

Reproduction and Growth

Flustra foliacea colonies are perennial; large colonies are several years old.

Young F.foliacea colonies are unilamellate and encrusting, when they reach a diameter of about an inch they give rise to erect bilamellate shoots by a process analogous to folding. These erect shoots grow and branch while the encrusting base spreads further over the support and gives rise to more erect shoots. In larger mature colonies there is a definite cycle of reproduction and growth. At the end of October vegetative growth ceases and embryos are present in the majority of the ooecia, the polyps soon degenerate and the colonies remain dormant. In late February the first swimming larvae are released, the polyps begin to regenerate and vegetative growth begins. During March and early April the majority of the larvae are released and vegetative growth quickens, the polyps are fully regenerated and active, and a well marked growth zone is present at the tip of the branch. This growth zone consists of rows of developing zooids and at the end of April when growth is in full swing consists of about 5-6 rows of partly differentiated zooecia, growth continues but becomes slower until October when the growth zone has returned to its dormant winter level of ½-1 row of incomplete zooecia. The eggs first become visible in August and begin to enter the ooecia in early October. By the end of October all vegetative growth has ceased and the eggs have all passed into the ooecia.

Observations suggest that the lines which cross the branches at intervals are a result of the annual cessation of growth from October to February. The interval between two of these lines represents a year's growth; these lines could probably be used to determine the age of the _Flustra foliacea_ colonies.

The embryos of _F.foliacea_ are usually produced in patches or zones on the frond where they give the area a distinct pinkish-orange appearance. The large larvae are orange (Munsell 7.5 YR 8/6).

Geographical Distribution

Murman Coast, White Sea, N.E.Atlantic to Biscay (Marcus 1940); Adriatic (Gautier, 1962).

SECURIFLUSTRA SECURIFRONS (Pallas)

(Hincks (1880), I.o.M.M.F.(1937) and Marcus (1940) as _Flustra_)

Distribution

Previously recorded from Manx waters but not collected in present survey.

Geographical Distribution

Arctic Ocean; N.Atlantic south to Newfoundland, Spain and Mediterranean.

HINCKSINA FLUSTROIDES (Hincks)

(Hincks (1880) and I.o.M.M.F. (1937) as *Membranipora*.)

Distribution

Previously unrecorded from the Isle of Man. Three specimens of *Hincksina flustroides* were dredged on shells in area C and one 6.2 ml (9.9 km) at S.19°E of the Chicken Rock in 37 f. (69 m).

Reproduction

Ooecia were absent in colonies collected in April and May but both eggs and embryos were present in February 1962 and November 1961.

Embryos are yellowish-orange in colour, rather more yellow than those of *Flustra foliacea* (Munsell colour not assessed).

Geographical Distribution

British Coasts, south to Morocco; Madeira; Mediterranean (Gautier 1962).

FAMILY MICROPORIDAE

MICROPORA CORIACEA (Johnston non Esper)

Distribution (See Map 25)

Micropora coriacea has not been collected on the shore or from soft grounds but is not uncommon in area C and the area stretching about ½ ml (.8 km) N-NW of C, elsewhere on coarse grounds it is rare. It occurs on a variety of dead shells and stones and grows most often on smooth surfaces.

Reproduction (See Fig. II 19 and II 20)

Embryos are present in small numbers from May to August. They are most abundant from September to April but even then less than 30% of the ooecia contain embryos.

Goutier (1962) recorded ooecia from May to September, and embryos in August in Mediterranean material.

Eggs, embryos and larvae are orange-red (Munsell colour: 2.5 YR 6/12).

Geographical Distribution

Widely distributed in the temperate Atlantic and Pacific, also Antarctic (Gautier, 1962).

FAMILY CELLARIIDAE

CELLARIA FISTULOSA Hincks (? non L.)

(Marcus (1940) as C.salicornia)

Distribution (See Map 26)

Cellaria fistulosa occurs on most coarse grounds and muddy sand grounds off the west coast of the island but is less common off the south coast. In few localities are more than 5 colonies present per 1000 cm^2 support surface area present but the conspicuous nature of the large, white colonies often give an impression of abundance greater than that indicated by quantitative samples. The finest colonies of this species, some larger than a clenched fist, occur on muddy sand grounds, e.g. areas A and E. Large colonies are usually attached to shells and stones by a network of stolons but tiny colonies of only a few zooids are rarely attached directly to these supports and are almost exclusively attached to hydroid stems and similar supports (some were found attached to the spines of Hermione hystrix (Savigny).). Stolons from the young colony attach it firmly to the hydroid and, by growing down the hydroid's stem, effect attachment to the support on which the hydroid is growing. Thus the primary support on which Cellaria fistulosa (and C.sinuosa) larvae settle is hydroid and only later by growth and secondary attachment do they fix to shells and stones. Large colonies occur most frequently on the outer surface of dead shells.

Reproduction (Fig. II 21)

Eggs were noted in November, December, February, April and May and embryos from September to May. The highest %s of colonies with embryos were noted in February, April and May.

Eggs and embryos are yellow (Munsell colour not assessed).

Geographical Distribution

Distributed in all warm, temperate and boreal waters but absent from Polar seas (Gautier, 1962).

CELLARIA SALICORNIOIDES Andouin (? Lamaroux)

(Hincks (1880) as C.johstoni)

Distribution

One colony was dredged in area A and one in area E.

Reproduction

Neither colony displayed any reproductive activity. Gautier

(1962) found ooecia all the year and embryos in May in Mediterranean material.

Geographical Distribution

Less widespread and of a more southerly distribution than C.fistulosa (Gautier 1962).

CELLARIA SINUOSA (Hassall)

Distribution (See Map 27)

The distribution of Cellaria sinuosa overlaps that of C.fistulosa but C.sinuosa is usually present in smaller numbers. Only in a very few samples were more than 5 colonies per 1000 cm^2 support surface area recorded. It is most abundant in areas A, B and E. Large colonies have been collected on a wide variety of dead shells and on stones but, like C.fistulosa, small colonies only occur on hydroids.

Reproduction (See Fig. II 22.)

Eggs were observed in September, November, December and a few in May; embryos were present from August to June. Larvae were released, in the laboratory, in March and April. The % of colonies with embryos was highest from November to March.

Gautier (1962) recorded ovicells from April to November and embryos in June and July.

C.sinuosa eggs are tiny and bright golden-yellow. Early embryos are golden yellow too, but during development become paler, the released larvae are almost white (Munsell 2.5 Y 8/4 and paler) with about 8 red 'eye-spots'. The ooeciferous zooecia occur principally in the distal half of each segment and when numerous embryos are present that part of the segment appears yellowish.

Geographical Distribution

E. Temperate Atlantic; Mediterranean (Gautier, 1962).

FAMILY SCRUPOCELLARIIDAE

SCRUPOCELLARIA SCRUPOSA (L.)

Distribution (See Map 28 and Table 17 and 18)

Widely distributed and locally common on coarse grounds but less so on softer, fine grounds, Sc. scruposa rarely forms over 5% of the total ectoprocts present in a sample. Sc. scruposa was not collected from the shore but was dredged in depths from 12 to 35 f. (22-64 m) and trawled on a Maia squinado in 41 f. (75 m) (See also p. 13). Sc. scruposa occurs on a wide variety of supports including dead shell, stones and hydroids. In area B it occurs on live Chlamys opercularis and in area C it is present on almost every shell. On dead shells (except Glycymeris glycymeris) Sc. scruposa is more common on the outer than the inner surface. The majority of the colonies on Glycymeris are primarily attached under the overhanging hinge of the shell.

Reproduction (See Fig. II 23)

Embryos were present from July to December with maximum % of colonies with embryos in September.

Roper (1913) recorded ooecia at Cullercoats in September and October. Gautier (1962) noted ooecia from May to September and embryo in March, May and September in Mediterranean specimens.

Eggs and embryos are red (Munsell colour not assessed).

Geographical Distribution

Temperate Atlantic from Iceland to Morocco and Madeira; Mediterranean; N.America?; Galapagos; New Zealand (Gautier 1962); Gulf of California (Steinbeck and Ricketts, 1941).

SCRUPOCELLARIA SCRUPEA Busk

Distribution

23 colonies were collected on dead shell: 15 from area C, 5 from B, 2 from A and 1 from E.

Reproduction

No activity was noted in Manx specimens. Gautier (1962) recorded ooecia from August to September, embryos in June, November and December and ancestrulae in May in Mediterranean collections.

Geographical Distribution

Temperate Boreal Atlantic; Azores; Cape Verde Is?, Mediterranean; Red Sea; Indian Ocean; Japan; E.Indies; Australia and New Zealand. (Gautier 1962)

SCRUPOCELLARIA REPTANS (L.)

Distribution (See Map 29)

Commonly present at about L.W.S.T. on most rocky shores where it occurs on rocks and stones and among red algal turfs, Scrupocellaria reptans, mixed with Crisidia cornuta and Crisia eburnea frequently forms a short turf covering extensive areas of vertical or overhanging rock at about L.W.S.T. and sub-littorally. Such turfs have been seen at Niarbyl, Fleshwick, Bradda Sker, Port Erin Breakwater, Perwick, etc. Sc.reptans is also very abundant on the alga Desmarestia aculeata (L.) Lamour.; over a hundred colonies may occur a single D.aculeata plant: D.aculeata plants being Sc.reptans were noted on sub-littoral rock when diving and were dredged regularily in area D, close to Bradda Hd. and on the landward side of area A. Sc.reptans also occurs in these and other localities with Sc.scruposa on dead shell and stones; Sc.scruposa is usually more abundant on dredged shell than Sc.reptans.

Reproduction (See Fig. II 24

Embryos were noted in all months except April and October, the highest % of colonies with embryos were recorded from June to August.

Roper (1913), at Cullercoats, found ooecia from May to October; Gautier (1962) working with Mediterranean material found ooecia in February, May, June, September and November and embryos in February, June and November.

Eggs, embryos and larvae are red.

Geographical Distribution

Temperate boreal Atlantic coasts of Europe; Mediterranean; Azores; Madeira (Gautier, 1962).

FAMILY BEANIDAE

BEANIA MIRABILIS Johnston

Distribution (See Map 30)

Beania mirabilis is widely distributed in small numbers on coarse grounds and slightly muddy sand grounds. It is generally absent from samples from muddy sand or mud grounds but has been dredged from the muddy ground D. In this investigation it has been dredged down to 37 f.(69 m) but Herdman (1893) records a colony from 60 f.(110 m) W. of Dalby. It is most common in depths from 15-25 f.(27-46 m). One colony was collected at E.L.W.S.T. on Port Erin Breakwater.

This species has been found straggling over a wide variety of supports and including most of the available species of dead shell, live *Anomia* spp., hydroids and the ectoprocts *Cellaria* spp., *Flustra foliacea*, *Lepralia foliacea*, *Eucratea loricata*, etc. It occurs in equal numbers on both surfaces of dead shell.

Reproduction

No data recorded.

Geographical Distribution

N.Scotland and Ireland represent this species' northern limit in Europe; it is recorded from all temperate and tropical seas. (Marcus 1940, Gautier 1962.)

FAMILY BICELLARIELLIDAE

BICELLARIELLA CILIATA (L.)

(Hincks (1880), I.o.M.M.F. (1937) as *Bicellaria*).

Distribution (See Map 31 and Table 19)

Bicellariella ciliata is scarce and of irregular occurrence in areas other than A, B and C where from 5-20 colonies may occur in a brdit sample. A single, small colony was collected among Crisia turf from L.W.S.T.L. at Calf Sound and a colony was trawled on a hydroid in 41 f.(75 m) but the species is most abundant, on coarse grounds with rich zoophyte growth, between 15 and 25 f. (37-47 m).

B. ciliata occurs on a variety of dead shells and zoophytes, e.g. the hydroids Hydrallmania falcata, Sertularella polyzonias, Abietinaria abietina and Sertularia spp., and the erect ectoprocts Cellaria spp., Scrupocellaria reptans and scruposa, Crisia spp., etc.

Reproduction (See Fig. II 25)

Bicellariella ciliata has two generations each year, one overwinters and its offspring are the rapid growing summer generation which in turn produces the next overwintering generation.

The 'summer' generation ceases to produce larvae by mid-November and soon dies and disintegrates. The offspring of the summer generation grow very slowly until about February-March when growth becomes very rapid. In 1961 the first embryos were noted in late March but in 1962 (a cold spring) they were observed until early May. By the end of May and during June the reproductive activity of this generation is at its peak and many larvae are being produced. The young colonies developing from these larvae grow rapidly and themselves bear embryos in the ooecia by August. The overwintering population has by this time almost ceased larval production and soon dies and disintegrates. Peak reproductive activity of the summer generation is in September but larvae are produced into November when this generation also dies. The offspring of the summer generation are the small overwintering colonies.

In large, reproducting colonies a zonation similar to that described by Correa (1948) in Bugula flabellata (Thompson) is present. From the tip of a branch towards the base of the colony are found successively (a) a zone of young zooecia without ooecia, (b) a zone of older zooids with empty ooecia, (c) a zone of zooecia with degenerating polyps and developing embryos in the ooecia, (d) a zone of zooecia with degenerated polyps, brown body, and fully developed larvae in the ooecia, (b') a zone with empty ooecia and regenerating polyps; and (c') zone of zooids with developing embryos in the ooecia, and so on. Each zone is usually only two or three zooecia long, but the complete zonation may be repeated once or twice in large colonies. This zonation gives insight into the pattern of life of the colony (See also *Bugula flabellata*, p. 56).

At Plymouth 'breeding' has been recorded in July and ooecia in March (P.M.F. 1957).

Geographical Distribution

Shetlands (not Faroes), British, Irish and other European coasts to the Mediterranean; W.Atlantic from islands of Canadian Arctic to Woods Hole; Red Sea; Indian Ocean; S.Australia; S.Africa.

FAMILY BUGULIDAE

BUGULA AVICULARIA (L.)

Distribution (See Map 32)

Bugula avicularia, like other British *Bugula* species, over-winters as inconspicuous colonies of only a few zooecia. The majority of deep-water samples were obtained in winter; during the summer, when *B.avicularia* colonies are large and conspicuous, samples were, for several unavoidable reasons, taken mainly at stations shallower than 25 f.(46 m). Thus the deep stations were not adequately sampled when *B.avicularia* was most conspicuous and therefore no reliable conclusions can be drawn about its distribution.

30% of the B.avicularia colonies collected were growing on erect zoophytes. The immediate support of the majority of colonies was an incrusting or nodular entoproct colony, the stolons of the B.avicularia ramifying among the zoids of the other colony.
(See also p. 55)

Reproduction (See Fig. II 26)

Embryos were noted from June to November; they are pale yellow. At Naples Lo Bianco (1908) recorded larvae in October.

Geographical Distribution

Widely distributed around British coasts (Ryland 1960), Shetlands, British and European coasts south to Morrocco, Madeira and Mediterranean; Red Sea; Loyalty Islands, Australia and New Zealand; Queen Charlotte Land; Vancouver; Panama (Marcus 1940).

BUGULA PLUMOSA (Pallas)

Distribution (See Map 32)

This species, like other Bugula species, over-winters as inconspicuous small colonies or as remnants of the previous summer's colonies. Any information on distribution is therefore likely to be biased by the preponderance of inshore hauls during the summer. A few small colonies were seen during the winter months, their polyps were active and not degenerated. Large colonies were collected from June to December, the majority were dredged in area B but colonies were collected from the mouth of Fleshwick Bay (10F; 18 m), area E and a few other localities. B.plumosa was not collected on the shore; the deepest colony was collected in 30 ft.(55 m) S.W. of the Chicken Rock.

B.plumosa occurs on a variety of supports including shells and stones, hydroids and Cellaria spp.

Reproduction

Empty ooecia were observed in August 1962 and embryos in September, October and December 1962.

Ryland (1960) recorded embryos from July to September and settlement in September in the Menai Straits.

Embryos are pale yellow.

Geographical Distribution

N.E.Atlantic from Norway to Madeira; Mediterranean; (Ryland 1960.)

BUGULA FLABELLATA (Thompson)

Distribution (See Map 32)

The overwintering ancestrulae, tiny colonies and tattered remnants of the previous summer's generation are fairly conspicuous and easily recognisable; it is unlikely that many were overlooked. Bugula flabellata is the commonest Bugula spp. in Manx waters and in summer regularly occurs in fair numbers (5-20 per bra%it sample) in dredged material from coarse inshore grounds such as A, B and C and from areas (e.g. area D, close to Bradda Head, etc.) where the bottom deposit is finer but there is still ample shell. B.flabellata is most abundant in areas A and C.

Superficially B.flabellata appears to grow on a wide variety of dead shells, stones, etc., but on close examination it is seen that every colony is attached (at least primarily) to another ectoproct colony usually of an incrusting species. The yellow stolons from the base of the B.flabellata colony ramify through the colony and at intervals put out erect shoots through the orifices of the 'host' colony. The first zooid of these erect shoots is ancestrula-like. The stolons ramify through living colonies without causing the death of the colony but some of the individual zooids of the 'host' colony are usually killed. Porella concinna (Busk) and Celleporaria pumicosa (Hincks) are most frequently 'infected' but B.flabellata colonies have also been observed on colonies of Schizomavella auriculata (Hassall); Cellaria spp., etc.

Reproduction (See Fig. II 27)

B.flabellata overwinters as ancestrulae and small colonies, as ancestrula-like erect shoots from stolons and as tattered, broken stumps of large colonies (these last may regenerate either by normal zooecial growth at the end of branches or by ancestrula-like shoots from individual zooecia). Few large colonies were seen between November 1961 and January 1962 and none between January and March 1962. Growth is very slow from November to March but rapid growth begins in March and in 1962 the first sexually mature colonies were seen in early May (embryo production may occur earlier in warmer years). By the end of May many colonies are reproducing and the offspring of these colonies become the rapid-growing summer generation. The summer generation begins to produce embryos by mid-September and soon after this the overwintering generation ceases reproduction and degenerates. The summer generation produces larvae until the end of October when all reproductive activity ceases and large colonies begin to degenerate. The overwintering population is a mixture of the stumps and stolons of the 'summer' generation and the ancestrulae and small colonies developing from the larvae of the 'summer' generation.

During peak reproductive activity zones of zooecia and embryos in different states of development occur along the branches of *B.flabellata* colonies (See also *Bicellariella ciliata*, p.53). Correa (1948) described similar zones in *B.flabellata* from Brazil.

Eggs, embryos and larvae are bright golden-yellow.

Ryland (1960) noted embryos from August to October and settlement in September at Menai. Gautier (1962) recorded ooecia from April to June and in October and December and embryos in April, October and December in the Mediterranean.

Geographical Distribution

North Sea and coasts of Europe from Skagerrak to Portugal; Atlantic coast of Morocco; Mediterranean; Adriatic; Mauritius;

Brazil (Ryland, 1960).

<u>BUGULA CALATHUS</u> Norman

<u>Distribution</u>

Previously recorded (I.o.M.M.F.) from "off Bradda Hd.", <u>Bugula calathus</u> was not collected in the present survey.

<u>Geographical Distribution</u>

S.W.Britain, Spain; Mediterranean; Adriatic; S.Africa. (Gautier, 1962).

<u>BUGULA TURBINATA</u> Alder

<u>Distribution</u>

One colony was dredged in area A and another from D. A fine specimen was collected while diving at Port Erin Breakwater. <u>Bugula turbinata</u> has been recorded from 33 f.(60 m), 6 miles W. of the Chicken Rock (I.o.M.M.F. 2nd edn.).

<u>Reproduction</u>

Embryos were present in September, October and November.

Rylands (1960) recorded embryos in Welsh specimens in August and September. Lo Bianco (1909) found larvae at Naples from February to May. Gautier (1962) found ooecia in September, November and December and embryos in September in Mediterranean material.

<u>Distribution</u>

Widely distributed in southern parts of British Isles, French coasts; Mediterranean and Adriatic (Ryland 1960).

<u>FAMILY CRIBRILINIDAE</u>

<u>MEMBRANIPORELLA NITIDA</u> (Johnston)

<u>Distribution</u> (See Map 33)

<u>Membraniporella nitida</u> is largely a species of the littoral

and immediate sub-littoral regions. It occurs on stones at L.W.S.T. on most rocky shores and on stones collected by diving in shallow water (20-30 ft: 11-18 m). It does, however, occur in some offshore areas, particularly where there is considerable water movement, e.g. area C, and has been dredged down to 30 f.(55 m).

Reproduction (See Fig. II 28 and II 30)

Eggs and embryos are present throughout the year with peak reproductive activity in 1961-62 in August-September.

Roper (1913) at Cullercoats, recorded ooecia in October and Gautier (1962) in the Mediterranean recorded ooecia from May to September and embryos from March to June.

Embryos and eggs are yellowish-orange (Munsell colour 2.5 - 7.5 YR $\frac{6-7}{8-10}$).

Geographical Distribution

European Atlantic coasts from Shetlands to Madeira and Mediterranean (Marcus 1940).

CRIBRILINA PUNCTATA (Hassall)

(Gautier 1962 as *Collarina*)

Distribution

Cribilina punctata is one of the commonest ectoprocts on Manx rocky shores. Where there is thick algal cover *C.punctata* occurs under stones from the upper *Ascophyllum*-zone to below low-water mark. A few colonies were dredged on shallow grounds, e.g. areas A and D.

Reproduction (See Fig. II 30 & 31)

During the period May 1961 to May 1962 embryos were present from August 1961 to May 1962. Very few embryos were seen in September 1961 and May 1962. Reproductive activity reached its peak from November to February.

Gautier (1962) found ooecia in Mediterranean material in April and October; Roper (1913) recorded ooecia at Cullercoats in April, May, August and December.

Eggs, embryos and larvae are red (Munsell colour 10.0R-25YR $\frac{5-7}{8-10}$).

Geographical Distribution

Temperate boreal Atlantic; Mediterranean; Arctico; Adelie Land; Indian Ocean (Marcus 1940).

CRIBRILINA ANNULATA (Fabricius)

Distribution

Very few colonies of C.annulata were collected, the majority were dredged in area C but some came from area B and one from 26 f.(48 m) at 5.7 ml (9.1 km), N.W. of the Sound. All were on the inner surface of dead shell.

Reproduction

Embryos were seen in July and November. Many of the fertile zooecia are raised above the rest of the colony.

Embryos are orange (Munsell colour 5.0 YR 6/10)

PUELLINA GATTYAE (Busk)

(Hincks (1880) and I.o.M.M.F.(1937) as Cribrilina)

Distribution

Not collected in this investigation. Recorded by Herdman (1896) from 16-20 f.(30-37 m) E. of the Calf Sound.

Geographical Distribution

Mediterranean (Gautier, 1962).

CRIBRILARIA RADIATA (Moll.)

(Hincks (1880) and I.o.M.M.F. (1937) as Cribrilina, Marcus (1940) as Colletosia.)

Cribrilina radiata is most abundant in area C and the western part of area B. It has also been dredged in small numbers in area A and from other, scattered localities on coarse grounds between 15 and 25 f.(27-46 m). Var α Hincks was dredged on two occasions, from 1 to 1½ mls (1.6 and 2.4 km) about S.E. of Spanish Hd. (Map 34)

41 of 45 specimens examined were growing on the inner surface of dead shells (mainly Chlamys opercularis and Glycymeris glycymeris)

Reproduction (See Fig.II 32)

Embryos were noted throughout the year. The highest % of colonies with embryos was noted in summer and the lowest from February to April (1962).

Gautier (1962) recorded ooecia in all months and embryos and larvae in January, July, November and December.

Embryos are dull orange-red.

Geographical distribution

E.Atlantic from Shetland to Cape Verde Islands; W.Atlantic from Georgia and Brazil; Azores; Tristan da Cunha; Mediterranean; Red Sea; Indo-Pacific Ocean from Japan to Zanzibar, Hawaii, Australia, Solomon and Loyalty Is., Tahiti and New Zealand. American W.Coast from Queen Charlotte's Land to Vancouver, Galapagos Is. (Marcus 1940).

FIGULARIA FIGULARIS (Johnston)

(Hincks (1880) as Cribrilina)

Distribution (See Map 35)

Previously unrecorded from Manx waters, F.figularis has a very limited distribution: it has been collected in small numbers from area C and the western part of B; It has also been dredged in areas A and D and two colonies were dredged S.W. of the Chickens.

The majority of colonies grow on the inner surface of dead shells.

Reproduction (See Fig.II 33 and 34)

No sample was examined in February and only very small samples were obtained in May, June and July. Embryos and eggs were present in all months in which samples were examined except July. No definite period of peak reproductive activity can be distinguished.

Gautier (1962) found ooecia throughout the year, embryos and larvae from May to December and ancestrulae in April, May and June in the Mediterranean. Lo Bianco (1909) recorded eggs and larvae from Naples in June. Ooecia have been recorded at Plymouth in April (P.M.F. 1957).

Embryos are red-brown (Munsell colour. 10.0R - 2.5YR$\frac{6}{10-14}$); the larvae are yellower (5.0 YR - 7.5 YR 6/8).

Geographical Distribution

E. temperate Atlantic from England and Ireland to Azores (Gautier 1962).

FAMILY HIPPOTHOIDAE

HIPPOTHOA DIVARICATA Lamouroux.

Distribution (See Map 36)

Widely distributed on shelly, fairly coarse grounds under 25 f.(46 m) but nowhere common, Hippothoa divaricata is often mixed with H.distans in samples; H.distans is usually the commoner. Two colonies of H.divaricata were collected on stones at E.L.W.S.T.

70% of H.divaricata colonies were growing on the inner surface of dead shells and 10% on stones.

Reproduction

Ooecia were observed from December to July; no embryos were seen.

Roper (1913) found ooecia in June, July and September at Cullercoats. Gautier (1962) found ooecia in May and June and embryos in June.

Geographical Distribution

Present in most cold and temperate seas but absent from tropical waters (Gautier 1962).

HIPPOTHOA DISTANS MacGillivray

(Hincks (1880) & I.o.M.M.F. (1937) and Gautier (1962) as H.flagellum)

Distribution (See Map 37 and Tables 10 and 21)

H.distans is much commoner than H.divaricata and has a much wider distribution, occurring throughout most of the area sampled. However, it rarely forms more than 5% of the ectoproct population or is present in numbers greater than 10 colonies per 1000 cm^2 surface area of shell and stone. H.distans occurs on most types of dead shell and on stones, it does not occur on algae or crustaceans and has not been collected on the shore. It is more common on smooth than on rough shell surfaces.

Reproduction (See Fig. II 35)

Embryos were noted from September to March and in May. The highest % of colonies with embryos was recorded in October 1961.

Gautier (1962) noted ooecia in April, May, June and October in Mediterranean.

Embryos are yellow.

Geographical Distribution

Cosmopolitan but living in the main in warmer waters than H.divaricata (Gautier 1962).

HIPPOTHOA HYALINA (L.)

(Hincks (1880), I.o.M.M.F. (1937) as *Schizoporella*)

Distribution (See Map 38)

On the shore *Hippothoa hyalina* is common below the lower *Fucus serratus*-zone and grows on algae, e.g. *Plumaria elegans*, *Laminaria* holdfasts, etc., and occasionally on the underside of stones. Sub-littorally it is abundant on *Laminaria saccharina* fronds; several hundred colonies may be present on a frond in the autumn. *L.saccharina* fronds with *H.hyalina* have been collected by diving and by dredging close to rocky shores, e.g. areas A and D, and close to Bradda Hd. etc. *H.hyalina* has also been dredged in small numbers on supports other than algae from various localities (particularly areas A, B and C) between 8 and 33 f.(15-60 m), about 70% of these colonies were growing on hydroids, a few on crab carapaces, one on a pycnogonid and the rest on dead shell. *H.hyalina* occurs much more frequently on smooth than rough shell surfaces.

Reproduction (Fig.II 36 and 37)

Large numbers of eggs, embryos and tiny colonies have been noted throughout the year.

Roper (1913) found ooecia in Northumberland specimens in September and October.

Eggs and embryos are yellow (Munsell colour 2.5-5.0 Y$\frac{8-9}{3-8}$).

Geographical Distribution

Cosmopolitan (Marcus 1940).

CHORIZOPORA BRONGNIARTI (Audouin)

Distribution (See Map 39 and Tables 20 + 21)

Chorizopora brongniarti is widely distributed on grounds where there is abundant dead shell. In some localities, e.g. around and including area C, more than 20 colonies per 1000 cm^2 surface area of support have been noted but only in a few localities are more than 5% of the ectoproct colonies of this species. C.brongniarti has been collected on the shore at Port Erin Breakwater and at the Sound and has been dredged in 37 f.(69 m).

No C.brongniarti colonies were seen on algae or hydroids, and only 17 of 997 colonies were collected on stones, the remaining colonies occurred on shell. C.brongniarti is commoner on smooth inner shell surfaces than rough outer shell surfaces.

Reproduction (Fig.II 38 and 39)

Embryos and eggs were noted in all months. Reproductive activity is highest in late summer (September to Octcber) and lowest in Spring (February to June).

Gautier (1962) recorded ooecia, embryos and larvae in all months and ancestrulae in April and August in Mediterranean material.

Embryos are red (Munsell colour 9.0 R$\frac{5}{10}$)

Geographical Distribution

European coasts south of Shetland; Morocco, Canaries, Cape Verde Is.; Mediterranean; S.Africa; Australia; New Zealand; Galapagos Is. (Marcus 1940).

HAPLOPOMA GRANIFERUM (Johnston)

(Hincks (1880) and I.o.M.M.F.(1937) as Microporella impressa; See Ryland 1963.)

Distribution

Haplopoma graniferum occurs under stones and boulders from MTL to below low water mark on all rocky shores examined and is probably present on sub-littoral rock. It has not been observed on algae other than Laminaria holdfasts. It has not been dredged.

Reproduction (See Fig. II 40 and 41)

Embryos were noted throughout the year; numbers were highest from June to September in 1961 and declined until April 1962, beginning to increase again in May 1962.

Gautier (1962) recorded ooecia in May, August, November and December and embryos in May in Mediterranean specimens.

Embryos are pink (Munsell colour 2.5-5.0 R $\frac{7}{4}$) early in development but become paler later in development.

Geographical Distribution

Shetland Islands to Mediterranean (Ryland 1963).

FAMILY RETEPORIDAE

SCHIZOTHECA FISSA (Busk)

Distribution (See Map 40)

Not common; less than 100 specimens were collected throughout this investigation. All were dredged between 18 and 27 f.(33-49m); the majority were dredged at area C, some at A, B and E, and the rest from a few other scattered localities. One specimen was growing on stone, the rest on the inner surface of dead shells.

Reproduction

No samples were collected in May. Eggs were observed from June to October and in February, and embryos in the same months and in December.

Gautier (1962) found ooecia in February, April, May, October, November and December and embryos and larvae in February, May and December in the Mediterranean.

Embryos are red.

Geographical Distribution

E.Atlantic Coasts from Great Britain to Cape Verde Islands; Mediterranean.

SCHIZOTHECA DIVISA (Norman)

Distribution

Schizotheca divisa was recorded by Herdman (1896) from 16-20 f.(29-37 m), E. of the Calf Sound. It has not been re-collected in the Manx area.

Geographical Distribution

Ireland, Channel Islands (Hincks (1880)).

FAMILY ADEONIDAE

REPTADEONELLA VIOLACEA

(Hincks (1880) and I.o.M.M.F.(1937) as Microporella.)

Distribution (See Map 41 and Table 22)

Reptadeonella violacea has been dredged between 18 and 37 f. (33-69 m.) and is locally common in areas with stony bottom and strong tides, e.g. close to the Calf (including area C) and the south of the island.

R.violacea from large spreading colonies ranging from white to purple in colour. The colonies on stone are usually larger than those on shell. In area C about 6 colonies are present per 1000 cm^2 surface area of stone, but only 1 or 2 columns occur on a similar area of shell.

Reproduction

Very large embryos are brooded in gonozooecia which are slightly larger and have a wider aperture than normal zooecia. Embryos were noted in January, from March to June and in September.

Gautier (1962) recorded gonozooecia from May to September and ancestrulae in April in the Mediterranean.

Embryos are brownish (Munsell colour 7.5-10.0 YR$\frac{5}{10}$). The larvae have several red "eye-spots".

Geographical Distribution

E.Atlantic from Britain to Cape Verde Is.; Mediterranean; Brazil; E.Pacific from California and Mexico. (Gautier 1962). Australia; Florida, Porto Rico (Marcus 1940).

FAMILY ESCHARELLIDAE

ESCHARELLA IMMERSA (Fleming)

(Hincks (1880) and I.o.M.M.F. (1937) as Mucronella peachii)

Distribution (See Maps 42 & 43 and Tables 23 and 24)

Escharella immersa is one of the commonest ectoprocts in Manx waters. It is common on stones and boulders in the rocky sub-littoral and on all rocky shores below L.W.N.T. It is widely distributed offshore and occurred in almost every sample and was dredged down to 37 f.(69 m). E.immersa is most abundant on coarse grounds and least common on muddy grounds. In many localities over 25 (and in some over 100) colonies are present on each 1000 cm^2 surface area of support. In most samples E.immersa makes up over 5% of the total ectoprocts present and in some localities, e.g. the Modiolus bed, over 50% of the ectoprocts present are this species.

No E.immersa colonies were observed on algae and only 1 colony of over 10,000 collected was attached to a hydroid. E.immersa occurs on a variety of shells and on stones but is most common on smooth surfaces.

Reproduction (See Fig. II 42 and 43)

Embryos and eggs were present in all months; a slight peak in reproductive activity occurred from February to May.

Eggs, embryos and larvae vary from pink to red (Munsell colours 2.5 $R\frac{5}{6}$, 5.0$R\frac{7}{6}$, 7.5-8.0$R\frac{5}{12}$ and 10.0$R\frac{7}{4}$ were recorded on different occasions).

Geographical Distribution

Arctic Region; N.Atlantic to Mediterranean and Woods Hole; Pacific to Vancouver (Marcus 1960).

ESCHARELLA VENTRICOSA (Hassall)

(Hincks (1880), I.o.M.M.F. (1937) as Mucronella)

Distribution (See Maps 44 & 45 and Tables 23 and 25)

Escharella ventricosa is widely distributed but generally less common than E.immersa although it is more common than E.immersa in some samples from offshore muddy sand. It is most abundant on muddy sand grounds but in few samples did over 20 colonies per 1000 cm^2 support surface area occur and it rarely makes up more than 10% of the total ectorpocts present. One specimen was collected on the shore; others were dredged in 37 f.(69 m).

Of 2500 colonies collected none were on algae, one was growing on a hydroid, and the remainder occurred on a variety of dead shell and on stones. E.ventricosa is more common on the inner than the outer surface of all dead shells except Cyprina islandica. Its apparent abundance on Chlamys opercularis is probably because C.opercularis is the dominant available support in the offshore areas where E.ventricosa is most abundant.

Reproduction (See Fig.II 44 and 45)

Embryos are present in all months; the highest percentages of colonies with embryos occur from November to March. Small colonies have been observed in all months.

Gautier (1962) found ooecia in August, September and November and ancestrulae in May in the Mediterranean.

Eggs and embryos are white.

Geographical Distribution

Similar to that of E.immersa.

ESCHARELLA VARIOLOSA (Johnston)

(Hincks (1880) and I.o.M.M.F. (1937) as Mucronella)

Distribution (See Map 46 and Tables 23 and 25)

Escharella variolosa is less common than the previous two species but is present on most of the shelly grounds around the south of the Island. E.variolosa is rarely present in numbers above 5 colonies per 1000 cm^2 surface area of support. It is most common where there is abundant tidal movement, e.g. area C and the tideway around the south of the island. Absent from muddy and very muddy sand grounds and not collected on the shore it has been dredged from 20 to 37 f.(37-69 m) and occurs on shells and stones. It is more common on the inner than the outer surface of dead shells.

Reproduction (See Fig. II 46)

Eggs and embryos have been noted in all months.

Gautier (1962) found ooecia in May, June and July and embryos and larvae in June-July.

Eggs, embryos and larvae are orange-yellow (Munsell colours 2.5 YR $\frac{6-7}{10}$ and 10.0 YR $\frac{7}{10}$ were observed on different occasions).

Geographical Distribution

E.Greenland, Hebrides, British, French and N.Spanish coasts; Morocco and Cape Verde Is.; Mediterranean; Vancouver (Marcus 1940).

ESCHARELLA ABYSSICOLA (Norman)

(Hincks (1880) and I.o.M.M.F. (1937) as Mucronella)

Distribution

Recorded by Herdman (1896) from Port Erin, this species has not been recollected in Manx waters.

Geographical Distribution

Arctic Ocean; Faroes, Norwegian coast south to Biscay and the Azores; Maine.

FAMILY SCHIZOPORELLIDAE

SCHIZOPORELLA UNICORNIS (Johnston)

Distribution (See Map 67 and Tables 26)

Schizoporella unicornis is common on rocky shores and forms large patch-like colonies on the underside of stones and boulders from the Ascophyllum-zone to below L.W. It is not common offshore and is confined to areas of strong tidal current and abundant stone and shell, e.g. area C and close to the south of the island. It has been dredged down to 32 f.(58 m). Offshore specimens are ARE usually form anasta Hincks and occur on shells and stones. It is more common on the inner than the outer surface of dead shells.

Reproduction (See Fig.II 47 and 48)

Embryos have been observed from June to January. Peak reproductive activity appears to be in June-July.

Gautier (1962) recorded ooecia from August to October and embryos and larvae in October in the Mediterranean.

Embryos are orange-red.

Geographical Distribution

Arctic region; Atlantic temperate and boreal coasts; Mediterranean; Indo-Pacific Ocean from Zanzibar to Japan, California.

SCHIZOPORELLA CRISTATA Hincks

Distribution

Recorded by Herdman (1896) from 16-20 f.(29-37 m) E. of the Sound but not collected in this investigation.

Geographical Distribution

Hastings (Hincks 1880).

SCHIZOPORELLA DISCOIDEA (Busk)

Distribution

Five colonies of this species previously unrecorded from the Isle of Man were dredged during 1961-62. All were on the inner surface of dead shell. Two were dredged at B, two at C and one at E.

Reproduction

All five colonies bore eggs and embryos. They were collected in September (two colonies), October and November 1961 and January 1962.

Eggs and embryos are red.

Geographical Distribution

Madeira; Gulf of Gascony; Mediterrancan (Gautier 1962).

SCHIZOMAVELLA AURICULATA (Hassall)

(Hincks (1880) and I.o.M.M.F. (1937) as Schizoporella)

Distribution (See Map 48 and 49, and Tables 26 and 27)

Schizomavella auriculata was not collected on the shore but is common offshore occurring in most dredged samples between 10 and 37 f.(18-69 m) and being trawled in 45 f.(82 m). In many samples over 10 colonies per 1000 cm^2 support surface area were noted and where it is most common (muddy sand grounds) over 50 colonies per 1000 cm^2 surface area are present. Frequently more than 10% of the colonies in a sample are of this species and in some areas, e.g. 1-2 ml (1.6-3.2 km) N.W. of Bradda Hd., more than a fifth of the ectoproct colonies are of this species.

S. auriculata occurs on a wide range of supports including stone, coal, clinker, living epifaunic molluscs and most types of dead shell. One of over 6900 colonies examined was on Cellaria, no colonies were seen on hydroids or algae. It grows on both rough and smooth surfaces and shows no ordered preference for the inner or outer surfaces of dead shell in terms of texture.

Reproduction (See Fig.II, 49 & 50)

Embryos and eggs were noted in all months with an obvious peak in reproductive activity between September and March.

Gautier (1962) recorded embryos throughout the year and ancestrulae in May and June in Mediterranean collections.

Eggs and embryos are brownish-red. Embryos become less red during development (Munsell assessments range from 2.5 YR$\frac{6}{14}$ to 10.0 YR$\frac{6}{10}$).

Geographical Distribution

Arctic Region and N.Atlantic coasts south to Madeira, Cape Verde Is. and Florida; Mediterranean; British Columbia to California; Korea and Japan to New Zealand.

SCHIZOMAVELLA LINEARIS (Hassall)

(Hincks (1880) and I.o.M.M.F. (1937) as *Schizoporella*.)

Distribution (See Map 50 and Table 26 and 27)

Schizomavella linearis is neither as common nor as widely distributed as *S.auriculata* but it occurs in moderate numbers over much of the area sampled. It is not uncommon on the underside of stones at E.L.W.S.T. on rocky shores and has been dredged down to 37 f.(69 m). More than 5 colonies per 1000 cm^2 support surface area were recorded from only a few samples and it rarely forms over 5% of the ectroprocts in a sample.

S.linearis occurs on stones, dead shell, etc. but has not been observed on algae or erect zoophytes. It is more frequently present on the inner than the outer surface of dead shell.

Reproduction (See Fig.II 51 and 52)

Eggs and embryos were seen in all months; reproductive activity is low during the colder months (February to April).

Gautier (1962) saw ooecia in all months and embryos and larvae from March to November in the Mediterranean.

Eggs, embryos and larvae vary in colour from red to orange-red (Munsell colours 2.5 $YR\frac{6}{12}$, 2.5 $YR\frac{6}{8}$, 5.0 $YR\frac{6}{8}$, 7.5 $R\frac{6}{10}$, and 10.0 $R\frac{6}{10}$ were recorded on different occasions).

Geographical Distribution

Arctic Region, European coasts to Morocco and Mediterranean; American W.coast from Vancouver and California; Indian Ocean (Marcus, 1940).

ESCHARINA SPINIFERA (Johnston)

(Hincks (1880) and I.o.M.M.F. (1937) as *Schizoporella*)

Distribution

Escharina spiniferum is abundant below the mid-*Fucus serratus* zone on rocky shores and extends on to shallow sub-littoral rock. It occurs on the underside of boulders and stones, on rock surfaces under algal cover and on *Laminaria* holdfats. Two colonies were dredged, one at 4.6 ml (7.3 km) S 79°W of the Chickens (depth 27 f. (49 m)) and the other 1.5 ml (2.4 km) N 79°W of the Chickens (depth 22 f.(40 m)).

Reproduction (See Fig.II 53 and 54)

E.spinifera has a well marked breeding season. Reproduction begins in January and reaches a peak in May and June and ceases by the end of July.

Eggs, embryos and larvae are orange-red. Late embryos and larvae are paler than early stages and have red 'eye-spots'. (Munsell colour: early stages: 10.0 R - 2.5 $YR\frac{6}{10}$; later stages: 2.5 - 5.0 $YR\frac{7}{10}$).

Geographical Distribution

Faroes to N.France; Adriatic (Marcus 1940).

ESCHARINA JOHNSTON (Quelch)

(I.o.M.M.F. (1939) as *Schizoporella*, Hincks (1880) as *Schizoporella simplex*.)

Distribution

Seventeen Escharina johnstoni colonies were dredged in this investigation: 11 from area C, 1 from B, 4 from localities close to C and one from 3.2 ml (5.1 km) at 44°S of E of Thousla Beacon (depth 27 f.(49 m)). 14 were growing on dead shell, the rest on stones.

Reproduction

Embryos were noted in January 1962 and May 1961; colonies observed in June, October, November 1960 and February, March and December 1961 were without embryos.

Geographical Distribution

Shetland, Irish, Scottish and English coats (Hincks 1880).

ESCHARINA VULGARIS (Moll)

(Hincks (1880) and I.o.M.M.F. (1937) as Schizoporella)

Distribution

11 colonies of Escharina vulgaris were dredged at area C and a further 18 from other scattered localities on shelly grounds. The deepest specimen was dredged in 35 f.(64 m). All were growing either on dead shells or on stones.

Reproduction

Embryos and eggs were present in colonies collected in March, June, August and December 1961, a colony seen in April 1960 had empty ooecia and colonies collected in January, March and May 1962 displayed no reproductive activity.

Gautier (1962) working on Mediterranean material found ooecia throughout the year and embryos and larvae in February, June, September, November and December and ancestrulae in January, May, August and September.

Embryos are orange-red.

Geographical Distribution

E.Atlantic coasts from Great Britain to Morocco, Azores and Cape Verde Is., Mediterranean; California (Gautier 1962).

ESCHARINA DUTERTREI (Audouin)

(Hincks (1880) and I.o.M.M.F. (1937) as *Mastigophora*)

Distribution

Less than 10 colonies were dredged in this survey; the deepest in 23 f.(42 m).

Reproduction

A colony collected in January had empty ooecia.

Geographical Distribution

Madeira; Azores; Mediterranean; Red Sea; Japan; Chile. (Gautier, 1962).

ESCHARINA ALDERI (Busk)

(Hincks (1880) and I.o.M.M.F. (1937) as *Schizoporella*)

Distribution

Herdman (1847) recorded this species from "off the west coast"; it was not collected during the present survey.

Geographical Distribution

Spitzbergen, N.Norway to Belgium and Ireland (Marcus 1940).

HERENTIA HYNDMANNI (Johnston)

(Hincks (1880) as *Mastigophora*)

Distribution

4 *Herentia hyndmanni* colonies were dredged on dead shells during this survey. It has not been previously recorded in Manx waters.

Reproduction

Eggs and embryos were noted in November 1961. Colonies examined in January and February 1960 and August 1961 displayed no reproductive activity.

Gautier (1962) found ooecia from May to November and saw embryos and larvae in July in Mediterranean specimens.

Eggs and embryos are pale red.

Geographical Distribution

Temperate boreal Atlantic; Mediterranean; S.Africa. (Gautier 1962)

FAMILY HIPPOPORINIDAE

HIPPOPORINA PERTUSA (Esper)

(Hincks (1880) and I.o.M.M.F. (1937) as *Lepralia*)

Distribution (See Map 51 and Table)

Hippoporina pertusa is widely distributed on muddy sand grounds and on muddy grounds where supports are available. It is nowhere abundant; more than 5 colonies per 1000 cm^2 support surface area occur in very few samples. It is rarely present on coarse grounds.

H.pertusa does not occur on zoophytes or algae; it has been collected on stones and a variety of shells. It occurs on both inner and outer shell surfaces.

Reproduction (See Fig. II 55 and 56)

Embryos occur throughout the year.

Gautier (1962) recorded ooecia from November to January and from June to August and embryos in the same months with the exception of August.

Embryos are bright orange. (Munsell colours 1.0 YR $\frac{6}{14}$ and $\frac{6}{10}$, 2.5 YR $\frac{6}{12}$ and 5.0 YR $\frac{7}{10}$ were recorded on different occasions.

Geographical Distribution

Appears to be cosmopolitan but some records are subject to query (Gautier 1962).

HIPPOPORIDRA EDAX (Busk)

(Hincks (1880) and I.o.M.N.F. (1937) as *Lepralia*)

Distribution

2 colonies of *Hippoporidra edax* were collected in this survey, one from 16f.(29 m) off Bradda Hd. and from area A. Both were on small gastropod shells containing the hermit crab *Pagurus cuanensis* (Thompson).

Reproduction

Embryos were noted in March 1963.

Embryos are orange-yellow (Munsell colour 3.0-5.0 YR 7/10).

Geographical Distribution

Florida (Hincks 1880).

CRYPTOSULA PALLASIANA (Moll)

(Hincks (1880) and I.o.M.N.F. (1937) as '*Lepralia*')

Distribution

Cryptosula pallasiana is common on most rocky shores from M.T. to below low water mark. It forms extensive sheet-like colonies on rock under thick algal cover, under stones, boulders and overhangs and in crevices. A few colonies were noted on algae. The I.o.M.N.F. 1962 records this species from 22 f.(40 m) but it was not dredged in the present survey.

Reproduction

No reproductive activity was seen.

Geographical Distribution

Atlantic Coasts from Bergen and Shetland to Madeira and Morocco and from Mt.Desert to Woods Hole, Pacific coast of America from Mexico and Alaska (Marcus (1940) and Gautier (1962)).

"LEPRALIA" FOLIACEA (Ellis and Solander)

(Gautier (1962) as "Hippodiplosia")

Distribution

Colonies of "Lepralia" foliacea have been dredged on several occasions at area C and on one occasion each from areas A and B and in 27 f. (50 m) at 3.2 ml (5.1 km) E 44°S of Thousla Beacon. In the Faunal Collections of the Port Erin Laboratory are specimens of L.foliacea which became entangled in fishing lines at the Calf Sound.

Reproduction

Embryos were present in June and September 1961. Colonies examined in May, October, January and February did not have embryos.

Embryos are bright yellow.

Geographical Distribution

European coasts from Arctic-boreal to Morocco; Indian Ocean? Alaska? (Gautier 1962).

"LEPRALIA" ADPRESSA Busk

Distribution

2 colonies of this species were identified on a small stone collected 3.5 ml (5.6 km) S 1° E of the Chicken Rock (depth 37 f. (59m)).

Reproduction

1 colony "dredged" in November bore a few ooecia.

Geographical Distribution

Mediterranean; Mazatlan? Chile?? (Hincks (1880)).

FAMILY MICROPORELLIDAE

MICROPORELLA CILIATA (Pallas)

Distribution (See Map 52 and 53 and Table 21 a 30)

Microporella ciliata commonly occurs on boulders and stones, red algae and *Laminaria* holdfasts etc. both on the lower shore and in the rocky sub-littoral. It occurs in very great numbers on *Laminaria saccharina* fronds. *M. ciliata* is widely distributed offshore and has been dredged down to 37 f (69 m). It is very common on coarse grounds but becomes less common with increasing fineness of the bottom substrate. In many of the samples from coarse grounds over 10% (and in some samples over 25%) of the ectoproct colonies present are of this species. *M. ciliata* colonies occur on a wide variety of supports, e.g. dead and living molluscs, *Laminaria saccharina* fronds, etc. It is more frequent on smooth than on rough surfaces. Only 19 of about 8500 colonies examined were growing on hydroids.

Reproduction (Fig. II 57 and 58)

Eggs and embryos are present in all months. Over 20% of the well-grown colonies examined bore embryos in all months except December and January. Peak reproductive activity is from May to September.

Roper (1913) found ooecia at Cullercoats in May, July and September. Gautier (1962) found ooecia in all months, embryos and larvae in January, February, April and September and ancestrulae in June, July and November.

Eggs, embryos and larvae are red, larvae and well-developed embryos are more orange than earlier developmental stages (Munsell colours are 10.0 R$\frac{5}{10}$ and $\frac{5}{8}$ and 5.0 YR$\frac{7}{10}$ (early developmental stages) and 2.5 YR$\frac{6}{10}$, 7.5-10.0 YR$\frac{5-6}{10}$, and 10.0 YR$\frac{6}{8}$ (later stages).

Geographical Distribution

All areas except the Antarctic Ocean (Marcus 1940).

FENESTRULINA MALUSI (Audouin)

(Hincks (1880) and I.o.M.N.F. (1937) as Microporella)

Distribution (See Map 54 & 55 and Tables 29-30)

Fenestrulina malusi was not present in shore collections; it has been dredged between 7 and 37 f.(13-69 m). Offshore it is abundant on coarse shelly grounds and is present in smaller numbers over most of the rest of the area sampled. It is least common on muddy grounds. In some samples, particularly those from shelly grounds between 20 and 30 f.(37-55 m), more than a tenth of the ectoproct colonies are F.malusi. F.malusi grows chiefly on shell: in area C (where over 50 colonies occur on each 1000 cm^2 support surface area), 16 colonies were noted, per 1000 cm^2 stone while over 100 colonies were present per 1000 cm^2 inner surface of dead shell. F.malusi is much more common on smooth than rough shell surfaces. Two of over 7000 colonies collected were growing on hydroids; none on algae.

Reproduction (See Fig.II 59 and 60)

Eggs and embryos were noted in all months. Reproductive activity was high from June to October but very few eggs and embryos were present between January and May.

Gautier (1962) found ooecia, embryos and larvae in all months and ancestrulae in January, April and May in Mediterranean collections.

On one occasion a larva was watched while it emerged from its egg-membrane. The fully-developed larva, still in its membrane, was removed intact and undamaged from its ooecium. The larva was revolving inside the membrane by ciliary activity. A tiny hole appeared in the wall of the membrane and a few cilia poked through it. The larva revolved until the powerful cilia of the vibratile organ protruded through the hole. These stout cilia made strong beating movements against the membrane which appeared to become softened and more flexible and finally split along about half its length. The larvae then retracted into the remnants of the membrane, revolved on its axis and then left the membrane and swam away. The process of emergence took place in about 30 seconds.

Eggs, embryos and larvae are golden-yellow in colour. Larvae are slightly paler than embryos and have four red "eye-spots" (Munsell colours 2.5 YR $\frac{5}{10}$ (early stages), and 10.0 YR$\frac{6-7}{8-10}$ (late developmental stages and larvae).

Geographical Distribution

Cosmopolitan but not continuously distributed (Gautier 1962).

FAMILY SMITTINIDAE

PALMICELLARIA SKENEI (Ellis and Solander)

Distribution (See Map.56)

About 150 colonies of this species were collected during this investigation, the great majority from muddy sand grounds. *Palmicellaria skenei* was dredged between 18 and 35 f.(33-60 m). 94 of 143 colonies growing on shell were on the outer surface.

Reproduction

The ooecia in this species are lightly calcified and delicate. Embryos were noted in September, December and April. Colonies without embryos were seen in August, September and November and from January to July.

Embryos are orange (Munsell colour 7.5 YR $\frac{6}{8}$)

Geographical Distribution

Arctic Ocean and Atlantic coasts south to Morocco and Maine; Mediterranean; Azores; Brazil. (Marcus 1940)

PORELLA CONCINNA (Busk)

Distribution (See Maps 57 & 58, and Table 31-32)

Porella concinna is widely distributed and particularly common on shelly muddy sand grounds. In a few samples over 25 colonies per 1000 cm^2 support surface area were noted; more than 5 colonies per 1000 cm^2 occurred in many samples. In a number of samples over 10% of the ectoproct colonies are P.concinna.

P.concinna grows on both rough and smooth surfaces of shells and on stones. One colony out of over 5000 examined was growing on a zoophyte (Cellaria fistulosa).

Reproduction (See Fig.II 61 & 62)

Embryos are present throughout the year.

Breeding has been recorded at Plymouth in July (P.M.F.1957) and at Cullercoats Roper (1913) found ooecia in July. Gautier (1962) found ooecia, embryos and larvae in January, March and April and ooecia only in June and September in Mediterranean collections.

Embryos are red (Munsell colours 7.5 $R\frac{5}{12}$, 10.0 $R\frac{6}{14}$, 2.0 $YR\frac{5}{10}$ were recorded on separate occasions).

Geographical Distribution

Arctic Region; Atlantic coast south to Madeira, Morrocco and Woods Hole; Mediterranean; Pacific coasts from Queen Charlotte's Land to Vancouver and Japan to S.Australia (Marcus 1940).

PORELLA COMPRESSA (Sowerby)

Distribution

Porella compressa was recorded by Lomas (1886) from south of Spanish Hd. A single, large but dead colony was dredged during the present survey in 30 f.(55 m) 2.2 ml (3.5 km) S 23°W from the Chicken Rock.

Geographical Distribution

Arctic Region; Iceland, Faroes, and European coasts to Biscay; Mediterranean; Hudston Str. (Marcus 1940).

PORELLA MINUTA (Norman)

Distribution

Herdman (1896) recorded Porella minuta from 16-20 f(29-37 m) E. of Calf Sound. Two colonies were identified during the present survey. Both were on Chlamys opercularis shells; one was dredged at area C and the other in 20 f at 1.2 ml (1.9 km) N 10°W of Thousla Beacon.

Reproduction

Both colonies, collected in December 1960 and February 1961, bore a few embryos.

Gautier (1962) recorded ooecia in March, June, August and September in Mediterranean specimens.

Embryos are brownish-red (Munsell colour 7.5-10.0 YR$\frac{7}{10}$).

Geographical Distribution

British Coasts; Mediterranean (Gautier 1962).

SMITTINA LANDSBOROVI (Johnston)

(Hincks (1880) and I.o.M.M.F. (1937) as Smittia)

Distribution (See Map 59)

Only about 100 colonies of Smittina landsborovi were identified in this survey. The majority were dredged in areas A, B, C and E but specimens were collected at a few other localities. Most were on dead shell but some were on living Chlamys opercularis Anomia spp.

Reproduction

Embryos were noted in January, March, October and November. Colonies without embryos were examined in June, September, December and February.

Gautier (1962) found ooecia, embryos and larvae in February, April and September and ooecia only in March in Mediterranean material.

Embryos are orange red.

Geographical Distribution

Appears to be cosmopolitan but a general revision of its distribution is necessary (Gautier 1962).

SMITTINA CHEILOSTOMATA (Monzoni)

(Hincks (1880) and I.o.M.M.F. (1937) as *Smittia*)

Distribution (See Map 59)

Smittina cheilostomata is sparsely distributed over shelly grounds, particularly where there is considerable tidal movement. 54 of 63 examined were growing on the smooth inner surface of dead shells.

Reproduction (See Fig.II 63 and 64)

Embryos were noted in all months except February, April and May; few were seen in January and March.

Reproducing colonies were found at Roscoff in September and October (Echalier and Prenant 1951). In Mediterranean specimens Gautier (1962) found ooecia, embryos and larvae in February and May and ooecia only in April, June and September.

Geographical Distribution

S. and W. Coasts of Britain; Roscoff; Mediterranean (Gautier 1962).

PARASMITTINA TRISPINOSA (Johnston)

(Hincks (1880) and I.o.M.M.F. (1937) as *Smittia*, Marcus 1940 and P.M.F. (1957) as *Smittina*.)

Distribution (See Map 60 and Tables 31 & 32)

Parasmittina trispinosa is not uncommon and is widely distributed off the S. of the Isle of Man. One colony was collected on the shore at Port Erin. It is most abundant on coarse, shelly grounds and uncommon on fine muddy grounds. Usually about 1% of the ectoproct colonies in a sample are P.trispinosa but in some localities its constitutes 5% of the ectoproct population.

P.trispinosa has not been collected on algae or zoophytes but occurs on a wide variety of shells and on stones. Colonies occur on both shell surfaces but are more numerous on the inner surface.

Colonies of P.trispinosa are yellow and frequently spread over areas greater than two square inches.

Reproduction (See Fig.II 65 & 66)

Eggs and embryos have been observed in all months.

Ooecia have been recorded in Northumberland in July and October (Roper 1913) and breeding at Plymouth in July (P.M.F.1957).

Eggs and early embryos are bright red (Munsell colour 7.5 R$\frac{5}{10\text{-}12}$, later stages and larvae are more orange (Munsell 5.0 YR$\frac{7}{10}$), all intermediate shades have been seen. In late embryos the cilia appear bluish through the ooecial wall.

Geographical Distribution

In all seas except Arctic and Antarctic (Marcus 1940).

SMITTOIDEA RETICULATA (MacGillivray)

(Hincks (1880) and I.o.M.M.F. (1937) as Smittia, P.M.F. (1937) as Smittina)

Distribution (See Map 61)

The revised edition of the I.o.M.M.F. lists Smittoidea reticulata as "fairly common on shells off the south of the Island" but I identified only about 100 of many thousands of ectoprocts examined as this species. Specimens occurred in small numbers from scattered localities between 7 and 35 f (13-60 m) off the south of the Island. S.reticulata was dredged most frequently at area C and on the Modiolus bed.

32 of 65 colonies of recorded support were growing on living *Anomia* spp. shells (*Anomia* constitutes only a small proportion of the available support in all areas). *S.reticulata* also occurs on living *Modiolus modiolus* on the *Modiolus* bed south of Spanish Hd. The majority of the colonies not on *Anomia* or *Modiolus* were on the inner surface of dead shell.

Reproduction (Fig. II 67 & 68)

Embryos and eggs were observed in June, August, September, November and December; eggs but not embryos were seen in January and February and ooecia only in March and April.

In Mediterranean collections Gautier (1962) found ooecia throughout the year and embryos and larvae from March to September.

Embryos are orange-red.

Geographical Distribution

E.Atlantic from Arctic to Cape Verde Is.; Mediterranean; Pacific coasts of N.America (Gautier 1962).

FAMILY PHYLACTELLIDAE

PHYLACTELLA COLLARIS (Norman)

Distribution (See Map 62)

60 *Phylactella collaris* colonies were dredged at scattered localities (including areas A, B, C, D and E) between 7 and 35 f. (13-64 m). All the colonies were on dead shell, the majority on the inner surface.

Reproduction

Samples were not obtained in January, April, or October; colonies with empty ooecia only were seen in February, March, May and July. Embryos were noted in June, August, September, November and December.

Embryos are orange-red.

Geographical Distribution

British Isles (Hincks 1880)

PHYLACTELLA LABROSA (Busk)

Distribution (See Map (2)

Of 35 Phylactella labrosa colonies identified over 20 were dredged in area B and the remainder from a few other, scattered localities. All were growing on the inner surface of dead shells.

Reproduction

Colonies displaying no reproductive activity were collected in January and February 1960, colonies with ooecia only were noted in December 1959, June and July 1960, and February 1961; a colony with embryos was dredged in August 1961.

Embryos are yellow.

Geographical Distribution

British Isles (Hincks 1880)

LAGENIPORA LEPRALIOIDES (Norman)

(Hincks (1880) and I.o.M.M.F. (1937) as L.socialis)

Distribution

Recorded from 16-20 f.(29-37 f) E. of the Calf Sound by Herdman (1896), this species has not been recollected in Manx waters.

Geographical Distribution

Boreal temperate E.Atlantic, Mediterranean (Gautier 1962).

FAMILY EXOCHELLIDAE

ESCHAROIDES COCCINEUS (Abildgaard)

(Hincks (1880) and I.o.M.M.F. (1937) as Mucronella)

Distribution (See Map 63)

Escharoides coccineus is a common species from the *Fucus serratus* zone to below low-water mark. It is abundant on the lower shore at Calf Sound and on parts of Port Erin Breakwater. *E.coccineus* forms extensive colonies over permanently damp rock surfaces such as the undersides of overhangs, under dense algal cover, under stones and boulders and on *Laminaria* holdfasts. It occurs on sub-littoral rock and has been dredged from a few localities on coarse shelly or stony grounds. The majority of dredged colonies were in samples from areas A, B and C. 70% of dredged colonies were on the smooth inner surface of dead shells.

Reproduction (See Fig.II, 69 & 70)

Embryos were observed in all months, few are present between September and January. Peak reproductive activity in 1961-62 was in July 1961 when over 40% of the colonies observed bore embryos. A large number of ooecia are present in most colonies but at no time did over 25% of them contain embryos.

In Mediterranean material Gautier (1962) found ooecia, embryos and larvae from May to November, only ooecia in January and ancestrulae in July and November.

Embryos are maroon (Munsell colours from 3.0 R$\frac{3}{8}$ for early developmental stages to 10.0R-2.5 YR$\frac{4}{8}$ for later stages and larvae were noted).

Geographical Distribution

E.Atlantic from Madeira to Spitzbergen (Gautier 1962).

FAMILY UMBONULIDAE

UMBONULA LITTORALIS Hastings

(Hincks (1880) and I.o.M.M.F. (1937) as *U.verrucosa*)

Distribution

Miller (I.o.M.M.F. 2nd edition, in prep.) records this species as "common on stones at L.W.S.T. around the south of the Island." During the present survey U.littoralis was occasionally present but never common in collections from between the lower Ascophyllum-zone and low-water mark on rocky shores.

Reproduction

In U.littoralis the embryos develop not in ooecia but within the parent zooecium. Up to 5 embryos occur together in a fertile zooecium.

Colonies with embryos were collected in February, September, October and November. Colonies without embryos were collected in January and from March to July.

Embryos are present at Plymouth in August (P.M.F. 1957).

Embryos are red.

Geographical Distribution

British Isles, Hardanger Fjord; Bergen (Hastings 1944).

UMBONULA ARCTICA (M.Sars)

(Hincks (1880) as Mucronella pavonella, Marcus (1940) as Discopora)

Distribution

Umbonula arctica has been previously recorded from off Port Erin (I.o.M.M.F. 2nd edition, in prep.) but was not collected in present survey.

Geographical Distribution

Arctic Region, Atlantic coasts south to Yorkshire and Woods Hole; In Pacific from Kurile Is., Queen Charlottes Land and Vancouver.

FAMILY CELLEPORARIIDAE

CELLEPORARIA DICHOTOMA (Hincks).

(Hincks (1880), I.o.M.M.F. (1937) and Marcus (1940) as Cellepora, Gautier (1962) as Harmerella.)

Distribution (See Map 64)

Celleporania dichotoma grows almost exclusively on hydroids; a very few colonies were dredged on shells. C.dichotoma is common on hydroids (particularly Abietinaria abietina (L.), Sertularella polyzonias (L.) and Sertularia spp.) on muddy sand grounds between 25 and 35 f.(47-64 m). It is not common on coarse grounds. It is rarely dredged in areas B and C, more common in A (where the substrate is muddier) and common at E.

Reproduction (See Fig.II 71)

A high percentage of the larger colonies bore embryos in all months.

Working on Mediterranean material Gautier (1962) found embryos from September to December.

Embryos are orange-red (Munsell 2.5 YR$\frac{6}{10}$).

Geographical Distribution

Scandinavia to Morocco and the Azores; Mediterranean; Florida? Japan? (Gautier 1962).

CELLEPORARIA PUMICOSA (Hincks)

(Hincks (1880), I.o.M.M.F. as Cellepora, Gautier (1962) as "Cellepora")

Distribution (See Map 65 and Tables 33 & 34)

Contrary to the statement in the I.o.M.M.F. (revised edition, in prep.) that Celleporania pumicosa is "common on the stems of hydroids and algae off the south of the Island" I find that C.pumicosa is rarely present on hydroids. Of 372 dredged colonies examined 14 were on hydroids, 28 on stones and 330 on shell. Some colonies occur on living Chlamys opercularis, Modiolus modiolus and Anomia spp.. C.pumicosa occurs on both surfaces of dead shells. It is fairly common on inshore dredging grounds (particularly area A and close to Bradda Hd. but is less common further offshore. C.pumicosa is present on the underside of stones and on Laminaria holdfasts at about low-water mark on most rocky shores.

Reproduction (See Fig.II 72)

Embryos are present in all months but in very small numbers between January and June.

Gautier (1962) found embryos and larvae throughout the year but less frequently in the warmer months in the Mediterranean.

Embryos are red. (Munsell colour 10.0R - 2.5YR$\frac{5}{10}$).

Geographical Distribution

Temperate boreal E.Atlantic from Norway to Morocco; Mediterranean. (Marcus 1940.)

OMALOSECOSA RAMULOSA (L.)

(Hincks (1880) and Marcus (1940) as Cellepora)

Distribution

A fine Omalosecosa ramulosa colony growing on Cellaria sinuosa was dredged in 30 f.(55 m) off Port Erin. No accurate position for the haul is available because of poor visibility.

Geographical Distribution

E.Atlantic from Morocco to N.Norway; Mediterranean. (Gautier, 1962).

FAMILY OSTHIMOSIIDAE

OSTHIMOSIA AVICULARIS Hincks

(Hincks (1880), I.o.M.M.F. (1937) and Marcus (1940) as

Cellepora, Gautier (1962) as "*Schizmopora*".)

Distribution (See Map 66 and Tables 33 & 34)

Osthimosia avicularis is fairly common and widely distributed over most of the shelly grounds investigated but is rare in offshore muddy areas. It was not present in shore collections. About half the *O. avicularis* colonies collected were growing on hydroids (particularly *Abietinaria abietina* (L.), *Sertularella polyzonias* (L.), *Hydrallmania falcata* (L.) and *Sertularia* spp.). Several *O. avicularis* colonies often occur on a single hydroid stem. The other 50% of the colonies were on both rough and smooth shell surfaces and on stones.

Reproduction (See Fig. II 73)

Embryos were present in all months. The percentage of colonies bearing embryos was highest between September and May.

Gautier (1962) found embryos and larvae in January, February, June and October, and ancestrulae in May in Mediterranean collections.

Embryos are yellow (Munsell colour 10.0 YR $\frac{7-8}{6-10}$).

Geographical Distribution

Arctic Region, E. Canada and European coasts from Norway to Cape Verde Islands; Mediterranean.

OSTHIMOSIA ARMATA Hincks

(Hincks (1880), I.o.M.M.F. (1937), Marcus (1940) as *Cellepora*, Gautier (1962) as "*Schizmopora*".)

Distribution

Herdman (1900) recorded this species from 30 f. (55 m) at 6 ml (9.6 km) S.E. of the Calf. This is the only record of *O. armata* in Manx waters.

Geographical Distribution

North Sea to Morocco; Mediterranean (Gautier 1962).

CELLEPORINA COSTAZII (Audouin)

(Hincks (1880), I.o.M.M.F. (1937) as *Cellepora*, Marcus (1940) as *Siniopelta*, Gautier (1962) as *C.hassalli*)

Distribution (See Map 67).

Celleporina costazii is common under stones, on small red algae and on *Laminaria* holdfasts at the lower levels of most rocky shores, it is also common on shelly grounds around the south of the Island. Over 90% of the dredged colonies were growing on hydroids, e.g. *Halecium halecinum* (L.), *Sertularella polyzonias* (L.) *Diphasia* spp., *Sertularia* spp. etc., the remainder on shells and stones.

Reproduction (See Fig.II 74)

Embryos were observed throughout the year, the % of colonies with embryos is highest from June to November. Large numbers of tiny colonies were noted on hydroids in March 1962.

Gautier (1962) noted ooecia throughout the year, embryos and larvae in February, May and June and ancestrulae in January, March, April, May, December.

Embryos are deep red (Munsell colours 7.5R$\frac{3}{10}$ (early stages) to 10.0R$\frac{5}{10}$ (late stages and larvae) were recorded).

Geographical Distribution

Temperate boreal Atlantic; other records must be regarded with suspicion (Gautier 1962).

FAMILY ALCYONIDIIDAE

ALCYONIDIUM PARASITICUM (Fleming)

Distribution

Several *Alcyonidium parasiticum* colonies were found incrusting hydroids (including *Hydrallmania falcata* (L.)) growing on the upper valve of a live *Pecten maximus* (L.) dredged at area C (depth 23 f. (42m)).

Geographical Distribution

Spitzbergen to the Channel; Chesapeake Bay; (Bobin and Prenant, 1956).

ALCYONIDIUM HIRSUTUM (Fleming)

Distribution

Alcyonidium hirsutum is common on *Fucus serratus* on rocky shores around the south of the Island. It occurs, but less frequently, on other algae, e.g. *Chondrus crispus* Stackh. and *Gigartina stellata* Batt. etc. and rarely on stones. It was not dredged but was collected on algae from the shallow sub-littoral by diving.

Reproduction

A.hirsutum has a well-marked reproductive season between September and April. The first sign of reproductive activity is the presence of numerous sperms (visible under the low-power binocular microscope as white patches) in mid-September and October. By November numerous small eggs or embryos are present. Each fertile zooecium contains 4-8 embryos. By late November all large colonies bear embryos. The first larvae are released in late January and during February the majority of the larvae are released. During March and April only a very few embryos remain in the parent zooecia. No embryos are present after April.

On several occasions larvae were observed on the surface of the colony after the colony was exposed by the receding tide, when placed in sea-water the larvae swam away from the colony in the normal manner. Large numbers of *A.hirsutum* larvae settle on *Fucus serratus* during January and February. The majority of ancestrulae are attached close to but on each side of the centre of the frond. The small colonies grow slowly; by early April (about 2 months after settlement) the majority of young colonies had only 2 zooecia, a fortnight later some had reached the 4-zooecia stage and two weeks later some colonies were of 6 zooecia, after this growth accelerated. As growth continued many of the colonies became contiguous and fused; by the end of July most of the colonies were of this composite nature.

The embryos of A. hirsutum are white and when mature form a conspicuous ring around the orifice of the parent zooid.

Geographical Distribution

Arctic Seas and European coasts south to the Channel (Bobin and Prenant 1956).

ALCYONIDIUM MAMILLATUM Alder

Distribution (See Map 68)

Less than 50 colonies of Alcyonidium mamillatum were collected during this survey. The majority were dredged on shells at area C and a few other scattered localities on shelly grounds. A few colonies were collected on the shore at Port Erin and the Calf Sound.

Geographical Distribution

Arctic Seas; British and North Sea Coasts; American Atlantic coast south to Maine; Brazil; Japan; Vancouver (Marcus 1940).

ALCYONIDIUM ALBIDUM Alder

Distribution

Specimens probably of this species have been dredged in several localities including area E and 4½ ml (7.2 km) at S.57°W of the Chicken Rock.

Geographical Distribution

Arctic Region and European coasts south to Biscay (Marcus 1940).

ALCYONIDIUM GELATINOSUM (L.)

Distribution (See Map 68)

Miller (I.o.M.M.F. 2nd edition, in prep.) recorded this species from L.W.S.T. on Port Erin Breakwater; it was not collected between tide-marks during the present survey. *Alcyonidium gelatinosum* occurs at areas A and B and in a few other localities. The majority of colonies collected were less than 3" high but a fine colony over 6" in height was dredged on the *Modiolus* bed. *A. gelatinosum* was collected from about 30' below L.W. on Port Erin Breakwater by diving.

Reproduction

Sperm-filled zooecia (which appear whitish and opaque) were noted in September and October; from November to February all large colonies contain embryos which are released in March. No reproductive activity was noted from March to August.

Geographical Distribution

Arctic Region and coasts south to S.E.England, Woods Hole and Vancouver; Mediterranean; Natal; Magellan Str. (Marcus 1940).

ALCYONIDIUM VARIEGATUM Prouho

Distribution (See Map 68)

Previously recorded only from Banyuls, colonies of this species occur on shells off Port Erin. Specimens agree in all respects with the description in Bobin and Prenant (1956) and in the reproductive season can be recognised by their bright-orange embryos. *Alcyonidium variegatum* has been dredged most frequently in areas A, B and C but also occurs in other localities. It occurs on both surfaces of dead shells and occasionally on stones.

Reproduction (Fig.II 75)

Embryos have been seen from August to May. About 8 develop in each fertile zooecium and appear as bright orange areas of colour under the low-power binocular microscope.

Geographical Distribution

Banyuls (Prouho 1892).

ALCYONIDIUM POLYOUM (Hassall)

Distribution

This species has been collected on the shore inside the T-block of Port Erin Breakwater. It was not collected on other shores. At Port Erin it occurs on *Fucus serratus* fronds and near the base of *Ascophyllum nodosum* fronds. It is not common. The closely related *Alcyonidium mytili* is common on the stones of this shore.

Reproduction

Many embryos were present in February 1962) and few in August 1962.

Hastings (P.M.F.1957) found larvae at Plymouth in August. Bobin and Prenant (1956) record breeding between April and August in the Channel.

Embryos are white.

Geographical Distribution

The synonymy and confusion associated with this species do not allow any conclusions to be drawn about its distribution.

ALCYONIDIUM MYTILI Dalyell

Distribution (See Map 69)

Alcyonidium mytili is common on stones from below the *Ascophyllum*-zone on most rocky shores. It is particularly common at E.L.W.S.T. among stones on the outer side of the 'T-block' at Port Erin Breakwater. It is common offshore on coarse shelly and stony grounds, especially where the tide is strong, e.g. area C and in the tideway around the south of the Island. *A.mytili* incrusts shells and stones.

Reproduction (Fig.II 76)

Embryos have beeen noted between October and June (with the exception of January), very few embryos were seen in October and from March to May.

Embryos are pale pink (Munsell colour 10.0 RP $\frac{8.5}{2}$).

Geographical Distribution

The synonymy and confusion associated with this species do not allow any conclusions to be made about its distribution. Definitely recorded from Morlaix (Le Brozec 1955).

FAMILY ARACHNIDIIDAE

ARACHNIDIUM HIPPOTHOOIDES (Hincks)

Distribution (Map 70)

25 colonies of *Arachnidium hippothooides* were collected in this survey. The majority were on *Ascidiella* spp. and on dead shell dredged on muddy sand grounds between 25 and 30 f. (45-55 m). A few were dredged on coarse grounds. Specimens on ascidian tests are very similar to those on shells but the individual zooids tend to be smaller in colonies on ascidians.

Geographical Distribution

Greenland, Spitzbergen, Torbay, Pas-de-Calais, Bonifacio (Bobin and Prenant 1956).

ARACHNIDIUM FIBROSUM Hincks

Distribution

A large colony apparently of this species was found on a *Sabella* tube from 57 f (103 m) N.W. of the Calf.

Geographical Distribution

Rare on N.Sea coasts; Ceylon; Brazil (Bobin and Prenant 1956).

FAMILY FLUSTRELLIDAE

FLUSTRELLIDRA HISPIDA (Fabricius)

(Hincks (1880), I.o.M.M.F. (1937), Marcus (1940), Bobin and Prenant (1956) and P.M.F. (1957) as Flustrella)

Distribution

Very abundant below L.W.N.T. on all algae-bearing rocky shores the south of the Island. Flustrellidra hispida is most common on Fucus serratus but also occurs on other algae e.g. Chondrus crispus, Gigartina stellata, Corallina officinalis, Plumaria elegans, etc., and less frequently on stones.

Reproduction

Reproduction takes place during the colder months. Sperm-filled zooecia were noted in November and small eggs in February. From March to June all large colonies contain some embryos. Each fertile zooecium broods from 4 to 8 embryos. In 1961 larvae and ancestrulae were noted from March to June but in 1962 (a colder year) ancestrulae did not appear until April and continued into July when observations ceased.

Geographical Distribution

Arctic; Atlantic coasts to Channel and Woods Hole; Pacific coasts from Arctic to California; Mediterranean? (Marcus 1940).

FAMILY VESICULARIIDAE

VESICULARIA SPINOSA (L.)

Distribution

4 Vesicularia spinosa colonies were collected, one from area A, one from B, one from 2.1 ml (3.3 km) at N 23°W of Thousla (22 f.(40m))

and one from 11 ml (17.7 km) S.W. of the Chicken Rock (42-45 f (75-82 m)).

Geographical Distribution

English; Irish; Belgium and Channel coasts, Mediterranean. (Bobin and Prenant, 1956).

BOWERBANKIA PUSTULOSA (Ellis and Solander)

Distribution

During the present survey a colony of *Bowerbankia pustulosa* was dredged from area D (10 f.(18 m)).

Reproduction

Embryos were present in September.

Embryos are yellow.

Geographical Distribution

S.Britain, Channel coasts; Mediterranean; Zanzibar (Bobin and Prenant, 1956).

BOWERBANKIA IMBRICATA (Adams)

Distribution

The I.o.M.M.F. (revised edition, in prep.) includes offshore records of this species but it was only found in shore collections during the present survey. It occurs on most rocky shores with algal cover from the *Ascophyllum* zone to low-water mark. It occurs on stones and near the base of *Ascophyllum nodusum* fronds.

Reproduction

Embryos were noted from June to September.

At Plymouth reproduction has been noted in August (P.M.F. 1957).

Embryos are yellow.

Geographical Distribution

Arctic Region; European coasts south to N.France; Mediterranean; Caspian Sea; Japan; Queen Charlottes Land. (Marcus 1940).

BOWERBANKIA GRACILIS Leidy

(Hincks (1880), I.o.M.M.F. (1937), Marcus (1940) and P.M.F. (1957) as B.caudata.)

Distribution

Bowerbankia gracilis has been collected on the underside of stones from most of the rocky shores of the south of the I.o.M. and has been dredged in small numbers in areas A, B, C, D, E, close to Bradda Hd. (depth 18 f.(33 m)) and 9.4 ml(15 km) at S 57°W from the Chicken Rock (depth 30 f.(55 m)). B.gracilis occurs on the walls of the Marine Biological Station aquarium tanks.

Reproduction

Embryos were noted in June and November.

Embryos are pink.

Geographical Distribution

Arctic Region, Atlantic coasts south to Channel and Gulf of Mexico; Mediterranean (Bobin and Prenant 1956).

AMATHIA LENDIGERA L.

Distribution

A colony of Amathia lendigera was found on the carapace of a large Maia squinado (Herbst) trawled S.W. of the Chicken Rock in about 40 f.(73 m) by the fishing boat "Manx Maid" and brought to the laboratory on 25.3.61.

Geographical Distribution

English, Belgium and French coasts; Mediterranean; Adriatic; Atlantic as far south as S.Africa; Indian Ocean as far south as Australia (Bobin and Prenant)1956).

FAMILY BUSKIIDAE

BUSKIA NITENS Alder

Distribution

Buskia nitens is very inconspicuous and difficult to see unless a careful search is made for it. It occurs on zoophyte stems. The embryos are bright yellow and more conspicuous than the rest of the colony which probably results in more colonies being noticed during the reproductive season than at other times. Colonies were noted on zoophytes from areas A, B, C and E but B.nitens probably has a wider distribution than this. One small colony was noted on the spines of a Hermione hystrix (Savigny)

Reproduction

The embryos are usually brooded singly but a few individuals contained two embryos. Embryos were noted from August to November. Embryos are bright yellow.

Geographical Distribution

Arctic Seas; Baltic; N.Sea; English and Channel coasts; Mediterranean; Porto Rico; Brazil; Zanzibar; Alaska. (Bobin and Prenant 1956).

FAMILY VALKERIIDAE

VALKERIA UVA (L.)

Distribution

Valkeria uva is a common species on algae-covered rocky shores, e.g. Port Erin Breakwater, the shores at Calf Sound, and Perwick Bay. Most colonies were collected on *Plumaria elegans* but it does occur on *Corallina officinalis*, other small algae and (rarely) on stones. It has been collected from about M.T. to L.W.S.T.

Reproduction

Embryos were noted in August and September.

At Roscoff reproduction takes place between May and September (Bobin and Prenant 1956).

Embryos are colourless.

Geographical Distribution.

Kara Sea; All N.Atlantic from Arctic south to Mediterranean and New York; Red Sea; Zanzibar; Indian Ocean; China Sea (Bobin and Prenant 1956).

VALKERIA TREMULA (Hincks)

Introduction

Valkeria tremula was briefly described by Hincks (1862 and 1880) who recorded *V.tremula* from the Isle of Man, Ilfracombe and Salcombe Bay. The only other record of the species is by Hallez (1890) from the Pas de Calais. Several colonies were collected in the present survey and a description of the species is given below.

Description

The colony consists of long, thin (25-35 μ) stolons composed of a series of elongate (2-3 mm) kenozooids. Each elongate kenozooid is swollen (to 40-60 μ) at its distal end immediately proximal to its terminal septa. From one or both sides of this swelling a small kenozooid is budded off which gives rise either to a series of similar small kenozooids or to a further elongate stolon-like kenozooid. On the small kenozooids and (rarely) on the swollen tip of the stolon tiny kenozooids are borne on which the autozooids are borne singly. The autozooids are deciduous and after an autozooid is shed the tiny kenozooid remains; up to 5 such tiny kenozooids have been noted on a small kenozooid as evidence of the former presence of 5 autozooids. Only one autozooid (rarely two) occurs on a small kenozooid at any time.

The autozooids are small; 400-650 μ in length, and 170-220 μ in width when fully contracted and almost twice as long when the tentacles are expanded. For most of their length the autozooids are of uniform width, but from about 40-70 μ from the proximal end (the end nearest the attachment) they taper rapidly until they measure only about 20 μ in diameter and are circular in cross-section where they attach to the tiny kenozooid which bears them. This tapered part of the zooid has the appearance of a peduncle. The cuticle is variously grooved at about 40-70 μ from the point of attachment.

In certain states of contraction the usually circular orifice becomes quadrangular under the action of the two series of parietal muscles. The tentacle sheath is long and bears a well-developed "pleated collar" (the "setose operculum" of Hincks (1880)) which folds spirally during retraction and closes the lumen of the invaginated tentacle sheath. The cuticle and body wall are transparent. There are eight campylonemate tentacles (i.e. two tentacles are always deflected outwards when the tentacular crown is expanded). The gut is of the usual ctenostomatous type. There is no gizzard. The stomach walls are brown. The musculature is of the typical ctenostomatous type. The lophophore retractors are inserted at the base of the tentacles and to the body wall distal to the grooved area of cuticle and about 100-200 μ from the proximal end of the autozooid. The tentacle-sheath retractors are well-developed and attach about and slightly distal to the pleated collar and to the body wall. The parietal musculature is modified so that the entire autozooid can make slight movements in the vertical plane. In the distal part of the autozooid the parietal muscles are, as in most stoloniferous ctenostomes, perpendicular to the longitudinal axis of the autozooid but the proximal pair of groups of fibres are not perpendicular but oblique. The two proximal groups insert close together on the wall near the attachment of the lophophore retractor muscles but on the other wall they insert separately so that they form an oblique V-shape across the zooecial cavity. Near their most distal attachment the body wall is modified so that contraction of these proximal parietal fibres results in invagination of the cuticle and consequent alteration of the axis of the autozooid though about 40°. These movements were mentioned by Hincks and take place as follows: for expansion of the tentacular crown the parietal muscles contract and the increase in hydrostatic pressure in the body cavity begins the eversion of the tentacular sheath. At the same time the contraction of the modified proximal parietal muscles causes invagination of the cuticle and lifts the distal part of the autozooid through about 40° into a position more nearly perpendicular to the support. Further parietal muscle contraction results in complete eversion of the tentacle sheath and tentacular crown. Retraction of the tentacles takes place in two stages. Until the pleated collar is level with the orifice the lophophore retractors and the retractors of the tentacle sheath contract together, but at this point the lophophore retractors reach their minimum length and further retraction is performed by the tentacle sheath retractors alone. While the lophophore retractors are still effective in retraction no drooping of the autozooid takes place, but once they cease to be effective slow drooping towards the anal side begins. Often retraction is only partial and the autozooids do not droop. Thus erection is controlled by modified parietal muscles by invagination of the body wall and drooping is not under direct muscular control. This method of erection is similar to that described by Silen (1950) for *Mimosella bigeminata* Waters and differs from *Farrella repens* (Farre) where the muscles controlling movement are enclosed in the peduncle.

Distribution

9 colonies of *V.tremula* were collected. Specimens were dredged in areas A, B, C, D and E which indicates a wide distribution but *V.tremula* is an inconspicuous species and has not been observed in samples from other localities. Colonies were on shells and on the erect ectoprocts *Cellaria fistulosa* and *Scrupocellaria scruposa*.

Geographical Distribution

Salcombe, Ilfracombe, Pas de Calais (Hincks 1880, Hallez 1890).

FAMILY TRITICELLIDAE

TRITICELLA KORENI G.O.Sars

(I.o.M.M.F. (1937) as *T.boeki*)

Distribution

Triticella koreni has been collected only on crustaceans and is common on *Nephrops norvegicus* (L.), *Calocaris macandreae* Bell and *Goneplax rhomboides* (L.) in Manx waters. *N.norvegicus* and *C.macandreae* occur only on mud or very muddy sand; all the *G.rhomboides* examined were dredged on muddy sand. *N.norvegicus* and

C.macandreae are abundant on mud grounds deeper than 37-40 f (69-73 m). T.koreni was collected between 37 and 65 f (69-119 m).

Observations on Manx material support Silen's (1936) con--clusion that Triticella koreni and T.boeki are varieties of a single species. On C.macandreae in Manx waters the majority of zooids are koreni-type but a wide variety of zooid shapes were noted on N.norvegicus. Koreni-type zooids occur on the carapace while boeki-type zooids are most common on the chelae, the rostrum, eyes, mouthparts and bases of the walking legs. Colonies with both koreni and boeki zooids and zooids which could not be classed as either have often been seen. Tentacle number varies between 10 and 22. The zooids with low tentacle numbers are usually on the rostrum or the chelae and are almost circular in lateral contour. It is unlikely that they are merely young zooids. Developing zooids with 18 incipient tentacles have been seen and zooids having only 14 tentacles have been seen with large eggs in the body cavity.

Reproduction

T.koreni autozooids are hermaphrodite but mature products of both gonads are not commonly free together in the body cavity. Zooids packed with sperm have been noted and up to 50 eggs have been seen in the body cavity of a single zooid.

Eggs and sperm were noted in all months in which samples were collected. Very few zooids contained eggs or sperm in January, May and November and in these months only a few eggs were present; in October about 20% of the zooids contained up to 10 eggs (and in some cases up to 30); in August the majority of autozooids contained over 5 eggs, some over 10 and some up to 50.

Geographical Distribution

British Isles, Norway, North Sea, Cape Breton, Biscay, Portugal; Adriatic; Sea of Marmara; Japan (Bobin and Prenant 1956).

FAMILY NOLELLIDAE

NOLELLA DILATATA (Hincks)

(Hincks (1880) and I.o.M.M.F. (1937) as *Cylindroecium*.)

Distribution (See Map 71)

Nolella dilatata is widely distributed on shelly grounds but is absent from mud grounds and extremely stony grounds. It is fairly common in areas A, B and C but less abundant elsewhere.

N. dilatata occurs on a wide variety of dead shells and zoophytes; few colonies were seen on stones. It has been recorded on spider-crabs and living *Chlamys opercularis* shells.

Reproduction

When embryos are present the distal part of the autozooid is swollen and sometimes appears whitish; 3-4 small white embryos are present in each fertile zooid. Colonies were examined in all months; embryos were noted in November, December and January.

Geographical Distribution

Arctic Region; European Atlantic Coasts; Mediterranean; Red Sea; India; Brazil (Bobin and Prenant 1956).

NOLELLA PUSILLA (Hincks)

(Hincks (1880) and I.o.M.M.F. (1937) as *Cylindroecium*)

Distribution

Recorded in Manx waters only by Herdman (1901) from off Contrary Head.

Geographical Distribution

British Isles (Ryland 1958).

FAMILY HYPOPHORELLIDAE

HYPOPHORELLA EXPANSA Ehlers

Distribution

The stolons of Hypophorella expansa ramify between the lamellae of the membranous tubes of certain polychaetes, e.g. Chaetopterus variopedatus (Renier) and Lanice conchilega (Pallas) and the autozooids protrude the tentacular crown into the lumen of the tube by way of a small circular hole. Each autozooid bears a "rasp" with which it keeps its hole open. H.expansa is common in Manx waters. It occurred in about 80% of the Chaetopterus tubes examined. Chaetopterus tubes with H.expansa have been dredged from much of the offshore sand and muddy sand areas. Only one of several Lanice tubes searched contained H.expansa.

Reproduction

Many of the zooids of a colony dredged in October 1961 contained several tiny eggs.

Geographical Distribution

North Sea, Channel, Banyuls. (Babin and Prenant 1956).

FAMILY MIMOSELLIDAE

MIMOSELLA GRACILIS Hincks

Distribution

Recorded from the Isle of Man only by Lomas (1886) from between Port Erin and the Calf.

Geographical Distribution

British, French, Spanish and Portugese coasts; Mediterranean. (Bobin and Prenant 1956).

FAMILY PENETRANTIIDAE

PENETRANTIA CONCHARUM Silen

Distribution

Penetrantia concharum is very common in Manx waters and was found wherever dead shells were collected. It has not been found on the shore but has been collected between 6 and 40 f(11-73m). *P. concharum* occurs in a great variety of dead shells. Almost every dead *Chlamys opercularis* (L.) and *Pecten maximus* (L.) shell is infected and it is commonly present in dead shells of *Glycymeris glycymeris* (L.), *Modiolus modiolus* (L.), *Ostrea edulis* L., *Cyprina islandica* (L.), *Dosinia lupinus* (L.), *Venus casina* (L.), *V. fasciata* da Costa, *Lutraria lutraria* (L.), *Gari* spp., *Ensis* spp., dead and living *Anomia* spp. (the first record of this species from live molluscs) and dead *Aporrhais pes-pelecani* (L.), and *Buccinum undatum* (L.).

Reproduction

Very few gonozooids were seen, none contained embryos.

Geographical Distribution

Norway, Sweden (Silen 1947) and California (Soule 1950).

SECTION III : ON THE DISTRIBUTION OF THE ECTOPROCTS OF THE SOUTH OF THE ISLE OF MAN

Introduction

One of the aims of this investigation was to acquire information on the distribution and the factors influencing the distribution of marine ectoprocts around the south of the Isle of Man. A large area (See p. 5) including a wide range of environmental conditions was sampled both qualitatively and quantitatively (See p. 6). The data obtained allow conclusions to be drawn as to the relative importance of various environmental factors in the ecology of ectoprocts. The only other study of this type is that of Gautier (1962) on Mediterranean cheilostomatous ectoprocts, the majority of other works on regional ectoproct faunas are largely taxonomical, e.g. Hincks (1880) "British Marine Polyzoa", Marcus (1940) "Danmark's Fauna 46. Mosdyr (Bryozoa eller Polyzoa)", Bobin et Prenant (1956) "Faune de France 60 Bryozaires Pt I", Osburn (1940)"Bryozoa of Porto Rico", etc.

Factors influencing the distribution of Manx Ectoprocts

SALINITY

The salinity of the sea around the south of the Isle of Man varies between about 33 and 35 ‰ (Slinn 1961) and cannot be expected to affect the distribution of ectoprocts in the area. No rivers enter the sea within the area sampled and most of the streams enter the sea over sand or through gravel. Ectoprocts are absent from the beds of the few small streams which enter the sea over rock or boulder beaches.

Ectoprocts are usually scarce or absent in rock pools where salinity (as well as p.H. and temperature) may undergo large fluctuations.

No freshwater ectoprocts have been recorded from the Isle of Man.

TEMPERATURE

There are no abrupt temperature changes in Manx waters: the annual temperature range is not great (about 6–16°C in most years) and temperature differences between surface and bottom waters are small throughout the year (See Fig II 77) (Slinn 1962). It is unlikely that temperature has much effect on the distribution of sub-littoral ectoprocts (See p. 135 for discussion on temperature and reproduction). On the shore abrupt temperature changes do occur and temperature has important effects on desiccation, humidity etc. The majority of shore ectoprocts occur in situations e.g. under stones or thick algal cover, where drying out is unlikely, but some species e.g. Flustrellidra hispida which is abundant on Fucus serratus, appear to be able to withstand considerable desiccation.

HYDROGEN-ION CONCENTRATION (p.H.)

The p.H. of the open sea varies within narrow limits and seems unlikely to effect the local distribution of ectoprocts. In rock-pools fluctuations in p.H. can take place; ectoprocts are not common in rock pools.

LIGHT

Because it is an important factor in the distribution of algae light has large effects on the distribution of shallow-water ectoprocts. Large algae serve as supports for many ectoprocts while small unicellular algae are the principal food of shallow water ectoprocts (see below). In Manx waters the growth of large, attached algae takes place only in the upper 22 m. (Kain 1960) (rock has not yet been found at greater depths) and consequently ectoprocts growing only on algae are restricted to these shallow waters.

Hyman (1959) states that the food of ectoprocts "consists of minute organisms, mainly diatoms" but it is obvious that the availability of diatoms will decrease with increasing depth. Mare (1941) working off Plymouth found several abundant benthic diatom species at 77 m. depth but only one at 113 m. Micro-organisms other than diatoms e.g. flagellate and ciliate protozoa, must make up an increasing proportion of the diet of ectoprocts with increasing depth.

The larvae of many ectoprocts become photophobic prior to settlement but the larvae of some species are indifferent to light (Ryland 1960). Ryland worked mainly with shore species. Photophobic larval behaviour would take the larvae into areas of low light intensity e.g. under stones and overhangs, where desiccation would be less likely and survival of the colony enhanced. In sub-littoral species photophobic behaviour would result in the larva reaching the bottom where it might find a support on which to metamorphose.

<u>FOOD</u> (See above)

It seems unlikely that food is of great importance in controlling the distribution of Manx ectoprocts. Availability of food would limit any ectoproct feeding only on algae to areas where suitable algae were present.

<u>AVAILABILITY OF LARVAE AND DISPERSAL MECHANISMS</u>

All ectoprocts with the exception of <u>Monobryozoon</u> spp. are sedentary and grow attached to a fixed support. The larval phase in the life-history is important both as a dispersal mechanism and as a mechanism for finding new areas of suitable substrate. Ryland (1959) showed that the larvae of some algal incrusting species select particular algae on which to settle. The larvae of other ectoprocts probably select particular supports for settlement. The present investigation has indicated that many species are more or less limited to a particular type of support and are not distributed at random. No experimental work has been done but the hypothesis that this non-random distribution is due to larval selection is borne out by the observed fact that few ancestrulae or tiny colonies of any species were found on supports outside the normal distribution of their species. If the non-random distribution was due to post-settlement natural selection then ancestrulae should occur on all available supports.

Two widely different types of larvae are found in marine ectoprocts. The majority of species have a lecithotrophic, short-lived larva but a few species have the more complex cyphonautes larva. The lecithotrophic larvae have a brief swimming existence of from a few hours to a day (Grave 1930, Hyman 1959, Ryland 1959). It is not known if metamorphosis can be delayed in the absence of a suitable support. Lecithotrophic larvae cannot disperse over more than a small area during their free-swimming life. Competition for support is greatest in those areas where supports are most abundant. Where support is scarce species which produce either more larvae per colony or longer-living larvae (both methods increase the number of "larvae-hours" per colony) could be expected to be most successful. How far this is true is not known but some of the commonest species in the muddy sand area e.g. Tubilipora liliacea, Lichenopora hispida, Alderina imbellis, Schizomavella auriculata, Osthimosia avicularis etc., produce large numbers of larvae. The majority of species found in areas of scant support (i.e. muddy sand) also occur in areas of more abundant support where, as one might expect, they are often equally or more common, but some species are confined to areas of scant support and it must be assumed that factors other than availability of support are of importance in their distribution. Such factors could be special food requirements or depth preferences etc., but species confined to these areas must have appropriately high "larvae-hours" per colony to enable them to maintain themselves. Thus the number and length of life of the larvae of a species are of importance in determining the extent of its distribution.

Cyphonautes larvae are planktonic for a period of several weeks and during this time become dispersed over a wide area. This is illustrated by the occurrence of Membranipora membranacea cyphonautes in the middle of the Irish Sea (D. I. Williamson personal communication); adult M. membranacea grow only on Laminaria fronds. Cyphonautes, in keeping with their longer planktonic life are produced in large numbers, up to 50 eggs being produced by a single zooid but lecithotrophic, short-lived larvae are produced in smaller numbers.

DEPTH

There are a number of species confined to the shore and immediate sub-littoral but below tide-marks depth alone is of little importance: coarse grounds have dense ectoproct populations, and silty, muddy grounds with little hard support have sparse populations irrespective of depth. But depth does affect water movement and bottom substrate which are important factors in ectoproct distribution.

WATER MOVEMENT

Both tidal currents and wave-action have large effects on the sea-bottom around the Isle of Man. In localities of powerful currents or wave-action all but the largest boulders and stones are swept away to be deposited where the strength of the water movement diminishes. Particles are deposited according to density, size and shape. In the area sampled the strongest tides (up to 4 knots) occur around the Calf (particularly through the Calf Sound and around Chicken Rock) and in those areas clean rock surfaces and large boulders make up the bulk of the bottom material. In areas of little or no water-movement e.g. some miles to the west and north-west of the Calf, the bottom material is almost entirely a fine, sticky mud. Between these two extremes a great variety of bottom types exists.

The occurrence of shell beds consisting mainly of one species e.g. the *Chlamys* beds west of the Chicken and the *Glycymeris* beds at area C, can perhaps be explained by reference to tidal currents. Shells of a particular species are fairly uniform in size and shape and if carried along by a tidal current will be deposited together when the current falls below a particular strength. The presence of a shell bed of a particular species is not necessarily correlated with an abundance of living specimens of the same species: live *Chlamys opercularis* are not common on the beds of *Chlamys* shells nor are live *Glycymeris glycymeris* common at area C. The sorting of bottom deposits by water movement affects not only the distribution of dead shells but also plays a large part in determining the point of origin of the shells. The distribution of many molluscs is strongly correlated with bottom deposits e.g. Glycymeris glycymeris lives only in areas of fairly clean sand while Cyprina islandica is commonest in muddy sand areas. In areas of little water movement the dead shell will be from molluscs originally living in that locality but where powerful tidal currents are sorting dead shell, shells of a species may be present where living specimens do not occur.

In addition to its importance in determining the nature of the bottom in a particular locality water movement has two other important effects, (a) it keeps hard surfaces clean and available for colonization by sessile organisms and (b) it ensures that a steady flow of water is moving past sessile organisms and thus increases the efficiency of filter feeders by reducing the likelihood of them filtering the same water twice.

The amount of exposure to wave action is an important factor in the ecology of rocky shores. Very exposed steep shores have no algae above the Laminaria zone and no stones or boulders and hence no ectoprocts but sloping shores exposed to little wave action have thick algal cover and often numerous stones and boulders and usually have a rich ectoproct population (see p. 125).

SUPPORT

All ectoprocts, except Monobryozoon spp., require a support for settlement and attachment. Any hard, clean support, e.g. shells, stones, algae, hydroids, crustaceans, etc., is likely to be colonized by ectoprocts. Support is probably the primary factor in ectoproct distribution but is itself dependant on many other environmental factors. In the area investigated the abundance of supports suitable for ectoproct colonization decreases with increasing fineness of the bottom sediments. In areas of fine sediments and little water movement (muddy sand and mud grounds) any hard, inert, material soon becomes covered with silt but on coarse grounds materials are kept clean of silt by water movement and are extensively colonized by incrusting organisms. The areas richest in ectoprocts are those with coarse bottoms and abundant water movement; muddy sand and mud grounds have poor populations (See Map 2).

Each ectoproct species grows only on a more or less limited range of supports which vary from species to species. The range of supports on which a species will grow is reflected in its distribution. The ubiquitous Electra pilosa (see p.33) grows on a great variety of supports but the closely related Membranipora membranacea grows on a narrow range of supports and has a much more restricted distribution. The range of supports on which a species can grow is, in part, dependant on its growth habit. On the basis of growth pattern ectoprocts fall into three major groups: erect species, encrusting species and nodular species. In encrusting species all the zooids are in contact with the support. In erect species only one or a few zooids are in contact with supports. Nodular species have a basal encrusting layer of zooids on which several more layers grow in turn. Encrusting species can be divided into three sub-groups: (1) species in which the colonies spread over the support by stolons, or by rows of zooids placed end to end either singly or in small numbers; (2) species producing flat, more or less circular, colonies in which all the zooids except a few of the oldest produce larvae; (3) species producing flat more or less circular colonies in which none of the zooids of a large central area and only a small proportion of those outside it produce larvae. The range of supports available for successful colonization is reduced as the area of support required for growth to mature size increases. Erect species require only a very small attachment area and, together with nodular species, and spreading species requiring narrow strips of support and encrusting species in which the majority of zooids brood larvae, can successfully colonize small areas (i.e. about $\frac{1}{2}cm^2$). Encrusting species in which only a small proportion of the zooids ever produce larvae must colonize a larger area if they are to grow to mature size. The ectoproct species from the south of the Isle of Man are grouped according to growth pattern in Table 35 which also indicates the main features of the distribution of each species. These differences in pattern of growth and support requirements mean that more species can exploit a given area of support. A single piece of shell can be colonized by erect, spreading and encrusting species.

The various types of support available for ectoproct colonization in the area surveyed are discussed in turn below.

ALGAE as supports for ectoprocts

Algae only occur where there is sufficient light for photosynthesis. Encrusting red-algae were dredged down to 29 f. (53 m.) and erect algae to 18 f. (33 m.). Encrusting algae are rarely colonized by ectoprocts. Other algae can be divided into broad and narrow-fronded types. Broad-fronded types include fucoids, Laminarias and some other red, brown and green algae. Narrow-fronded algae are species such as Desmarestia aculeata Lamour, Halidrys siliquosa Lyngb., Chorda filum Lamour., Plumaria elegans Schm., Ceramium spp., etc. Laminaria holdfasts are treated separately below. Broad fronds allow successful colonization by incrusting ectoprocts and a number of species grow on broad-fronded algae. Stoloniferous spreading ectoprocts rarely occur on broad-fronded algae. Narrow-fronded algae rarely provide sufficient area for incrusting species to grow successfully and are colonized mainly by rampant and erect species. The ectoproct populations of Laminaria digitata holdfasts are similar to those of rocks and stones at the same shore level and consists mainly of incrusting species. The bulbous holdfasts of Saccorhiza polyschides rarely bear ectoprocts.

Experiments have shown (Ryland 1959, Crisp and Williams 1960) that the larvae of algal dwelling ectoprocts settle on particular algae when offered a choice. Frequently the preferences shown in the laboratory correspond with the field distribution of the species.

The ectoprocts occurring on algae are discussed below and listed in Table 36 .

No ectoprocts were found at higher levels on the shore than the Ascophyllum zone. On Ascophyllum nodosum itself colonies of Flustrellidra hispida are frequent and Bowerbankia imbricata occasional on the holdfast region and on the lower stipes. Flustrellidra hispida and Alcyonidium hirsutum are both extremely common on Fucus serratus but competition for support is partly reduced by differences in distribution: Fl.hispida is most abundant on the stipes and lower fronds while A.hirsutum is most common on the fronds. Fl.hispida larvae are produced later in the year than A.hirsutum larvae. Both ectoprocts also occur in smaller numbers on other algae. A few colonies of Alcyonidium polyoum have been collected on F.serratus and Electra pilosa and Membranipora membranacea also occur on this alga. Membranipora membranacea is abundant on Laminaria holdfasts particularly in summer (August - October). Colonies of Electra pilosa, Hippothoa hyalina, Alcyonidium hirsutum and Flustrellidra hispida frequently occur on broad-fronded red algae e.g. Chondrus crispus and Gigartina stellata and are less frequent on narrow-fronded algae e.g. Corallina spp., Plumaria elegans etc. Colonies of Crisidia cornuta, Crisia eburnea, Scruparia spp., Valkeria uva occur frequently on narrow-frond red algae and occasionally on broad-fronded species. Colonies of species such as Callopora lineata, Microporella ciliata and Celleporina costazii occasionally occur on littoral algae.

On Laminaria holdfasts the ectoprocts are those of neighbouring rocks and stones and include the species Tubulipora phalangea, Callopora lineata, Hippothoa hyalina, Schizomavella linearis, Celleporina costazii and several other species in smaller numbers.

The only sub-littoral algae examined were various Laminariales and Desmarestia aculeata. Scrupocellaria reptans is very common on D.aculeata and Electra pilosa sometimes occurs on this alga. Few ectoprocts have been recorded on Alaria esculenta or Saccorhiza polyschides but other Laminariales regularly bear ectoprocts. Membranipora membranacea is abundant on the fronds of Laminaria digitata and Laminaria hyperborea and is sometimes present on the fronds of L.saccharina. L.saccharina fronds commonly bear hundreds of ectoproct colonies which are more frequent in the concavities than on the convexities of the frond; the majority of these colonies are of Callopora lineata, Hippothoa hyalina and Microporella ciliata but colonies of Aetea sica, Bugula flabellata, Beania mirabilis and Cellopora craticula have also been noted on this alga. Electra pilosa encrusts many L.hyperborea stipes but ectoprocts do not occur on the stipes of other Laminariales. Holdfasts of L.hyperborea and L.digitata frequently bear numerous colonies of encrusting ectoprocts; Cellopora lineata, Hippothoa hyalina, Escharina spiniferum and Escharoides coccineus are the commonest species.

ZOOPHYTES as supports for ectoprocts

With the exception of Flustra spp. and "Lepralia" foliacea, erect zoophytes provide only a small surface area for ectoproct colonization. Few encrusting ectoprocts grow on zoophytes. Hydroids have nematocysts and ectoprocts have aviculariae and these offensive weapons probably deter prospecting ectoproct larvae. The hydroids most frequently colonized by ectoprocts e.g. Hydrallmania falcata, Sertularia polyzonias, Halecium halecinum etc. have a central stem devoid of polyps which is frequently the only part which bears ectoprocts. The lateral polyp-bearing branches of these hydroids and the central polyp-bearing stems of other hydroid species are rarely colonized but Celleporina costazii sometimes occurs on them. Bugula avicularia, with many large aviculariae is never colonized by other ectoprocts while Eucratea loricata which lacks aviculariae is frequently colonized.

Ectoprocts with erect, rampant and nodular colonies occur on zoophytes. Encrusting species rarely occur on zoophytes but Electra pilosa and Alcyonidium parasiticum are exceptions and incrust hydroid stems. Erect ectoprocts attach to the zoophytes by the primary zooid and by stolons and require very little surface area for colonization. Crisidia cornuta, Crisia spp., Bicellariella ciliata, Bugula spp., Scrupocellaria spp. are of this erect type. Cellaria fistulosa and C.sinuosa larvae settle on hydroids but by massive extension of stolons down the hydroid stem the Cellaria colony soon becomes attached to the support on which the hydroid is growing. Thus Cellaria spp. grows on hydroids for the early part of their life. Nodular species e.g. Celleporina costazii, Cellepораria dichotoma and Osthimosia avicularis etc. attach to the zoophyte (usually a hydroid) by a calcareous holdfast and do not have stolons. They either form nodular colonies of several layers of zooecia around the hydroid stem (e.g. O.avicularis) or give rise to erect shoots from a nodular base (C.dichotoma). Rampant, spreading ectoprocts in which the zooids are separated by stolons or stolon-like elongations of the zooid frequently occur on zoophytes. Most species of this type have the orifice raised on an erect elongation of the zooid (e.g. Aetea spp., Nolella dilatata)but some species (e.g. Buskia nitens) do not have the orifice raised in this way.

In general the ectoprocts which occur on zoophytes grow on suitable hydroids and ectoprocts alike but Cellepororaria dichotoma and Buskia nitens were recorded only on hydroids and Bugula flabellata colonies grew primarily on ectoprocts. The majority of ectoprocts growing on zoophytes also occur on shells but the reverse is not true.

The ectoprocts collected on zoophytes during the present survey are listed in Table 37 . Species marked with an asterisk are not typically present on zoophytes and are not discussed further. Growth on erect zoophytes raises an ectoproct above the level of the bottom and thus any species growing on zoophytes is feeding in the same water level as the large erect ectoproct species such as Eucratea loricata and Flustra foliacea. Groups I and II of Table 37 are not included in the following discussion.

Of the species in Group III, Cellaria fistulosa and Cellaria sinuosa grow on zoophytes for the early part of their life only (see p.47). The other species in Group III show differences in distribution which are likely to reduce competition. The distribution of Bugula avicularia and B.plumosa is not fully known but they appear to be commonest on coarse and shelly grounds. Widespread species, present wherever suitable supports occur, include Crisia eburnea, Scruparia spp., Beania mirabilis, Electra pilosa and Osthimosia avicularis. Species more or less confined to coarse and shelly grounds are Crisidia cornuta, Aetea anguina and Nolella dilatata. Species more or less confined to muddy sand are Tubulipora liliacea, Celleporaria dichotoma, Buskia nitens, Bowerbankia gracilis and Valkeria tremula. Spreading species rarely take up such a high proportion of the available support as to exclude other spreading species e.g. on coarse grounds Aetea anguina is often mixed with colonies of Nolella dilatata or Beania mirabilis and on muddier grounds Bowerbankia gracilis and Valkeria tremula colonies often intermingle. Closely related species usually show differences in distribution on the zoophyte reducing direct competition between them: Osthimosia avicularis grows only on the main axis of hydroids, Celleporania dichotoma grows mainly on lateral branches but close to the axis while Celleporina costazii grows on laterls branches away from the axis. Crisidia cornuta and Crisia eburnea both grow on hydroids on coarse grounds but C.eburnea tends to grow higher up the hydroid than C.cornuta.

Thus competition is reduced in zoophyte-inhabiting ectoprocts by differences in distribution and growth habit but nevertheless it is not uncommon to find a hydroid colony bearing colonies of up to ten ectpproct species.

SHELLS and STONES as supports for ectoprocts

Shells and stones are the principal supports colonized by ectoprocts in the Manx area. They generally provide sufficient surface area for the growth to mature size of all colony-types. The amount of shell and stone present increases with increasing coarseness of the bottom substrate. Shell and stone are absent from mud-grounds. Shell beds occur in most sand and coarse-gravel areas. Where tidal currents are very strong e.g. close to the Calf and to Chicken Rock, the only material dredged is stones and boulders. Stones are rarely dredged away from strong currents but the local scallop-fishermen state that when a scallop bed is fished intensively for some time "the stones come to the surface". Such stones are clean and without sessile organisms. On rocky shores and in the rocky sub-littoral stone is available for colonization but shell is almost absent.

The shells most frequently dredged are those of Chlamys opercularis, Pecten maximus, Glycymeris glycymeris, Cyprina islandica, Modiolus modiolus and Buccinum undatum with Venus spp., Gari spp., Dosinia lupinus etc. in smaller quantities. The proportions of the various shells in a sample varies from place to place (see p.116). Shells and stones are colonized by many ectoproct species. Usually more colonies are present on the inner than the outer surface of dead shells. Shells and stones are most densely colonized on coarse grounds (see p.124).

Shells of different species vary in physical characters. The degree of convexity and concavity varies from species to species and from part to part of the same shell. Most shells have smooth concave inner surfaces but Pecten and Chlamys shells have broad, smooth ribs on the inner surface. Texture of the outer shell surface varies greatly. There are many intermediate textures between the glass-smooth shell of Ensis spp. and the deep ridges of Venus casina or the hispid surface of Chlamys opercularis. The influences of surface texture on the distribution of individual species are noted in the account of each species. Table 40 summarizes these results. Some species e.g. Fenestrulina malusi and Escharella variolosa cannot tolerate rough surfaces while others e.g. Amphiblestrum flemingi occur most often on rough surfaces while still others e.g. Porella concinna are indifferent to surface texture. On Laminaria saccharina ectoprocts occur most frequently in the concavities of the frond, it may be that similar preferences for concavities and convexities are important in the distribution of shell and stone dwelling ectoprocts. Osthimosia avicularis and Bicellariella ciliata, species common on the convex stems of hydroids, when present on shells usually occur at the edge of the shell or at the apex of Pomatoceros triqueterr (L.) tubes. Shells with a well developed periostracum e.g. Ensis spp., Cyprina islandica and Modiolus modiolus are less frequently colonized by ectoprocts than are species without well-developed periostraca e.g. Chlamys opercularis and Pecten maximus.

Table 38 lists the species occurring on shells in the area examined. It does not include rare species. Species in Group I are infrequent on shells in the Manx area and are not considered in the following discussion.

Although many species grow on shell few are distributed throughout the area where shell is available and some have a very limited distribution. A number of shell-inhabiting species (including *Flustra foliacea*, *Figularia figularis*, *Cribrilaria radiata*, *Micropora coriacea* etc.) have been collected only at or near area C. *Palmicellaria skenei*, *Alderina imbellis*, *Hippoporina pertusa* and *Cellepcraria dichotoma* are found mainly in muddy sand areas. Of the remaining species the majority are most common in areas of abundant available support and considerable water movement such as area C. In such localities very dense populations are found and competition for support must be intense. Many more ectoprocts have been collected on *Glycymeris*, *Chlamys* and *Pecten* shells than on other types of shell but this is probably because they are the commonest shells in areas of powerful water currents. In muddy sand areas *Pecten* and *Chlamys* shells do not bear richer ectoproct populations than other shells.

On the *Modiolus modiolus* bed south of Spanish Head shell of other species is virtually absent. The ectoproct population of this bed is noteworthy for the paucity of species present. *Escharella immersa* is by the far the most abundant species. *Diplosolen obelia* and *Smittoidea reticulata* are more abundant on the *Modiolus* bed than elsewhere.

Living epifaunal molluscs are frequently colonized by ectoprocts. The lower valves of live *Chlamys opercularis* are more extensively colonized than those of *Pecten maximus*. This difference is presumably due to the different habits of the molluscs: *C.opercularis* is more active than *P.maximus* and does not dig into the substrate to the extent that the *Pecten* does. *Amphiblestrum flemingi* and an *Alcyonidium*? sp. are frequently present on the lower valves of living *C.opercularis*. Gastropods such as *Buccinum undatum* are frequently colonized by ectoprocts.

Shells are also colonized extensively by the boring ectoproct *Penetrantia concharum*. The activities of this ectoproct together with other shell-boring organisms e.g. the sponge *Cliona celata* (Grant) and *Phoronis* spp., are important factors in the breakdown of shells in the sea. *P.concharum* bores by phosphoric acid (Silen 1947).

Stones are in many ways similar to shells and offer extensive surface area for colonization by ectoprocts. On the shore the undersides of stones not fixed in sediments, the undersides of overhangs and rocks under thick algal cover afford shelter from desiccation and temperature extremes and are colonized by ectoprocts. Sub-littorally there is little ecological difference between stone and rock. Offshore grounds with abundant stone are of limited extent. Because of the sorting action of tidal currents all the stones in an area tend to be of a similar size. Stones are usually smooth and rounded but in some localities they are pitted with the tunnels of boring molluscs. Many stones in shallow water bear incrusting red algae which are rarely colonized by ectoprocts.

Small stones less than ½" diameter and small fragments of shells are colonized by only a few ectoproct species notably Pyripora catenularia and Hippothoa distans, the limited area available does not allow colonies of other species to reach mature size.

Ectoprocts occurring on rock and stone on the shore include the following (see also Table 35):

In the Ascophyllum zone: Cribrilina punctata, Schizomavella unicornis and Bowerbankia imbricata are common under stones with occasional colonies of Alcyonidium mytili, Bowerbankia gracilis and Valkeria uva.

Fucus serratus zone: Under stones the species from the Ascophyllum zone plus Caulorhamphus spiniferum, Callopora aurita, Haplopoma graniferum, Microporella ciliata, Escharella immersa and Escharina spiniferum. Cryptosula pallasiana and Umbonula littoralis occur both under stones and on permanently damp rock surfaces.

Laminaria digitata zone: All the above species with the exception of Cryptosula pallasiana have been collected in this zone. Escharoides coccineus is very common on rock faces and Scrupocellaria reptans, Crisidia cornuta and Crisia eburnea also occur on rock faces . Under stones most of the species noted in the F.serratus zone are common with the regular addition of Tubulipora phalangea, Lichenopora hispida, Electra pilosa, Callopora lineata, Schizomavella linearis, Hippothoa hyalina, Cellepora costazii and Celleporaria pumicosa, many other species are present in smaller numbers.

Table 39 lists the species growing on stone off the Isle of Man; it does not include shore records (see p.125). Species rarely present on stone (Group I of Table 39) will not be considered further. The majority of species growing on stones also occur on other supports. If sufficient support is available all the species in Group III form large, sheet-like colonies. "Lepralia" foliacea colonies consist of a large attachment area from which erect foliaceous sheets arise. None of the other species found principally on stones are erect. The larvae of Group III may have evolved a preference for stones as only stones provide sufficient surface area for the colonies to reach a large size. Groups II and III include the species likely to be present on dredged stones but they are not all equally abundant. The most abundant species on stones are Crisidia cornuta, Crisia denticulata, Lichenopora hispida, Tubulipora spp., Electra pilosa, Pyripora catenularia, Hippothoa divaricata, Microporella ciliata, Escharella immersa, Porella concinna, Reptadeonella vidacea, Schizomavella linearis and Alcyonidium mytili. In localities where species of Group III are very common they cover much of the stone surface and leave little space for other species.

ARTHROPODS and OTHER ORGANISMS as supports for ectoprocts

Ascidians, tube-worm opercula, crustaceans and even pycnogonids, have been collected with ectoprocts growing on them. The majority of these associations are outside the normal distribution of the ectoproct species involved and are probably of an accidental nature but there are a few examples which appear to indicate a genuine affinity on the part of an ectoproct for a particular, living, support. Most of the Arachnidium hippothooides colonies collected were on simple ascidians. Triticella koreni has been collected only on the crustaceans Calocaris macandreae, Nephrops norvegicus and Goneplax rhomboides. The association between T. koreni and these crustaceans is probably due to the restriction of T. koreni to mud grounds where crustaceans are the only available support. (In the Firth of Clyde T. koreni occurs on Chlamys septemradiata (Müller) from mud grounds (Allen 1953 as T. pedicellata). Hippoporidra edax has only been collected on small gastropods containing hermit crabs. A high % of shells containing large hermit crabs are colonized by Electra pilosa (See p. 33), but E.pilosa is a common species and occurs on other supports. The membranous tubes of Chaetopterus variopedatus are frequently colonized by Hypophorella expansa. Spider crabs frequently carry pieces of ectoproct attached to the carapace; the majority of these are probably placed there by the crab.

With the exception of the examples mentioned above any settlement of ectoprocts on unusual supports must be regarded as accidental. The majority of crustaceans have elaborate cleaning rituals (with the exception of spider crabs in which rituals having the opposite effect have evolved) which must reduce colonization by encrusting organisms but occasional ectoproct colonies do become established, particularly on the larger crustaceans. Species most usually found on crustaceans are those with a wide range of supports e.g. Lichenopora hispida, Electra pilosa, Schizomavella auriculata and S.linearis, Escharella immersa etc.

Geographical distribution of Manx ectoprocts and comparison with other areas.

In the present survey 130 of the 142 ectoproct species recorded from the Isle of Man were collected. 38 of the species recorded from the I.o.M. have not been recorded at Plymouth; 13 species recorded at Plymouth have not been collected in Manx waters. Data on the geographical distribution of Manx ectoprocts have been taken from Bobin and Prenant (1956), Canu and Bassler (1925, 1928, 1929), Echalier and Prenant (1951), Gautier (1962), Hastings (1941, 1944), Marcus (1926, 1940), Maturo (1957), Osburn (1923, 1940), Rogick (1956), Ryland (1956, 1958, 1963), Silen (1954), Soule (1954) etc. But for 8 species I have found no distribution data other than that in Hincks (1880) but Hincks' geographical data are regarded as unreliable by most authorities. Of the remaining 134 species, as far as I am aware, 6 are confined to the European Boreal, 51 have been recorded both from the Mediterranean region (including S.Spain and

N.W.Africa) and the Arctic region, 49 have been recorded from the Mediterranean and not the Arctic and 15 from the Arctic but not the Mediterranean. 13 species are recorded from other regions but not from the Mediterranean or the Arctic. Thus the ectoproct fauna of the south of the Isle of Man consists largely of Atlantic-Boreal and Widespread species with an excess of Mediterranean-Atlantic species over Arctic-Atlantic. This situation is similar to that found in the Manx nudibranchs by Miller (1959).

Ryland (1962) discusses the distribution of shore ectoprocts with particular reference to Welsh localities (Menai Straits and Anglesey, Milford Haven). He records 47 species including 10 (Electra crustulenta, Conopeum reticulum, Scrupocellaria scruposa, Bugula plumosa, B.fulva, B.flabellata, B.turbinata, Fenestrulina malusi, Cellepora (=Osthimosia) avicularis and Amathia lendigera) which I have not collected on Manx shores. All except Electra crustulenta and Bugula fulva have been collected in Manx waters. Filicrisia geniculata, Diplosolen obelia, Diastopora suborbicularis, Aetea sica, Beania mirabilis, Callopora aurita, Amphiblestrum flemingi, Membraniporella nitida, Chorhizopora brongniarti, Hippothoa divaricata, Haplopoma graniferum, Escharella ventricosa, Parasmittina trispinosa, Alcyonidium mammillatum and A.mytili are not mentioned by Ryland but have been collected on Manx shores. With the above exceptions the two surveys are in close agreement. The ectoprocts occurring on Manx shores are listed in Table 41 .

Knight-Jones and Jones (1955) sampled the ectoproct fauna of the sub-littoral rock off Bardsey. They recorded about 60 species. There is no reason to suppose that the ectoproct fauna of Manx sub-littoral rock is poorer than that of Bardsey.

Conclusion and Summary

Of the various factors in the marine environment around the south of the Isle of Man it appears that temperature, salinity, p.H. and depth have little direct effect on the distribution of ectoprocts. Light affects ectoproct distribution indirectly through its importance in controlling the distribution of algae which are important to ectoprocts as food and potential supports. The type and abundance of larvae may also affect ectoproct distribution. All ectoprocts except _Monobryozoon_ spp., require a support and the distribution of supports (which is dependant in turn on water movement) is the most important factor in ectoproct distribution. Supports can be divided into three main types, algae, zoophytes and shells and stones. Some ectoprocts can grow on most types of supports but the majority occur in a more or less limited range of supports. Supports of one sort or another suitable for ectoproct colonization are present throughout the area. Generally speaking any clean hard support can be colonized by one or another type or species of ectoproct.

Many species have a wide distribution in the area investigated but some species are confined to certain types of locality and can probably be regarded as 'indicator' species analogous to plankton 'indicator' species. Just as plankton 'indicators' indicate the presence of a particular type of water so these ectoproct species indicate the presence of particular bottom conditions.

Species found only in areas of very muddy sand and mud: _Triticella koreni_.

Species typical of muddy sand areas and less common elsewhere: _Alderina imbellis_, _Palmicellaria skenei_, _Escharella ventricosa_, _Cellaporaria dichotoma_, _Arachnidium hippothoöides_.

Species found mainly in areas of powerful water currents:

On shell: _Figularia figularis_, _Cribrilaria radiata_, _Micropora coriacea_.

On stone: "_Lepralia_" _foliacea_, _Pentadeonella violacea_, _Escharoides coccineus_, _Schizoporella unicornis_.

The majority of the other species (excluding those confined to the shore) are most abundant in areas of considerable water movement and abundant varied supports and become less common with decreasing water movement and amount of clean, hard, support. Because of this it is not possible to speak of ectoprocts in terms of different communities on different grounds. The majority of species are commonest in areas such as C which can loosely be described as areas 'best for ectoproct growth' and the distribution of each species through the rest of the area appears to depend on its toleranco of less favourable conditions. The distribution of Manx ectoprocts is summarized in Table 35

The Manx ectoproct fauna consists largely of Atlantic-Boreal and widely distributed species with an excess of Mediterranean-Atlantic species over Arctic Atlantic.

SECTION IV : ON THE REPRODUCTION OF THE ECTOPROCTS OF THE SOUTH OF THE ISLE OF MAN

Introduction

Data on the season of reproduction of marine ectoprocts have been collected from various sources: there are a few works concerened entirely with ectoproct larvae (Barrois 1877, Prouho 1892, Atkins 1955), studies on fouling (e.g. Coe and Allen 1937, Nair 1961, 1962) sometimes include data on the settlement of ectoprocts and some fauna lists include information on reproduction (Echalier and Prenant 1951, P.M.F. 1957). Marcus (1926) compiled a table including all the data available to him on the season of reproduction in European ectoprocts. However all these scattered sources do not give full or satisfactory data on the season of reproduction of the ectoprocts of any given area. The fullest study so far published is that of Gautier (1962) in which he gives information on the season of reproduction of 164 species (including 50 recorded for the I.o.M.) of Mediterranean ectoprocts but for 64 of these he gives information only on the occurrence of ooecia or ancestrulae. The presence of ooecia in a colony indicates that the colony is either about to reproduce or has reproduced. Only the presence of eggs or embryos can be taken as definite evidence of reproduction at a particular time. Gautier's data is not quantitative and his collections were made at several widely separated Mediterranean localities. So far as I am aware the present survey is the only one in which the reproduction of ectoprocts in a small area has been examined throughout the year. The aim of the investigation was to obtain as much information as possible in as quantitative a manner as possible on the reproduction of Manx ectoprocts. The methods used are described on p. 7 and the results obtained are given separately in Section II for each species in turn. The data obtained show that there is a wide variety of reproductive habits in the Manx ectoprocts.

Reproductive Habits

Manx ectoprocts can be classified on the basis of their reproductive habits as follows:

A. Non-brooding species e.g. Membranipora membranacea, Electra pilosa, Hypophorella expansa, Triticella koreni.

B. Brooding species

I Embryos brooded in unmodified zooecia.

(i) Embryos brooded singly e.g. Bowerbankia, Valkeria uva.

(ii) Embryos not brooded singly e.g. Cryptosula pallasiana, Umbonula littoralis, Alcyonidium spp., Flustrellidra hispida, Nolella dilatata.

II Embryos brooded in more or less modified zooids.

(i) Embryos brooded singly e.g. Reptadeonella violacea, Penetrantia concharum.

(ii) Embryos not brooded singly e.g. all Cyclostomata.

III Embryos brooded in ooecia.

(i) "Oöecia", membranous, evanescent e.g. Aetea spp., Eucratea loricata.

(ii) Ooecia calcareous, permanent e.g. most Cheilostomata.

The number of larvae produced by a species is correlated with the amount of protection given to the embryos and the type of larva produced. Brooding species with a short larval life produce fewer larvae than those species with larvae having a long planktonic life. Cyclostomatous ectoprocts produce large numbers of simple short-lived larvae in each ovicell. The number of ovicells in each cyclostome colony is usually small but varies from species to species.

The very large larvae of Reptadeonella violacea are produced in small numbers while species like Callopora lineata produce smaller larvae in larger numbers.

Table 43 shows that of 13 species typically common on the shore and less common elsewhere 8 brood their embryos internally. The distribution of these shore species indicates that internal brooding affords better embryo protection than brooding in ooecia. 7 of the 8 internally brooding species occur in localities where some desiccation might occasionally occur e.g. on algae or exposed rock faces, while the 5 typical shore species which brood the embryos in ooecia all occur in sites where desiccation is less likely e.g. under stones and overhangs. All the ooeciferous species which occur both on the shore and sub-littorally occur in sheltered positions on the shore. 8 internally brooding species occur sub-littorally (excluding cyclostomes).

Table 42 includes the 52 species (excluding cyclostomes and cyphonaute-producing species) for which full data on reproductive seasons have been obtained and shows the relationship between length of reproductive season, proportion of fertile zooids in a colony and distribution. The first point is that no typical shore species (i.e. species uncommon elsewhere) have long reproductive seasons. Many shore species compensate for their short reproductive season by producing large numbers of embryos in it. Fertile zooids of internally brooding species usually brood several embryos at once e.g. *Alcyonidium* spp., *Umbonula littoralis* etc. and in ooeciferous species most of the zooids are fertile and produce embryos. Equal numbers of species with high and low proportions of fertile zooids have a limited offshore distribution but the majority of widely distributed species have high proportions of fertile zooids in the colony. The largest category in Table 42 , those species with a high proportion of fertile zooids and a long reproductive season, include many of the most abundant Manx ectoprocts. The other large category, offshore species with many fertile zooids and a short reproductive season, also appear to 'compensate' for their short season by producing large numbers of larvae in it; this group includes common species such as *Bicellariella ciliata*, *Bugula* spp., *Cellepora pumicosa* etc.

Length of period of development

Silén (1945) found that the embryos of *Callopora dumerili* spend about a fortnight in the ooecium between passing from the body cavity into the ooecium and swimming away from the ooecium as a larva. No individual embryos were watched during the present investigation but in species with a well de-limited reproductive season the difference between the first appearance of the embryos and the first larvae gives an indication of development time. Data were obtained from several species. _Flustra foliacea_ eggs first pass into the ooecia in early October and the first larvae are released in late February. _Alcyonidium hirsutum_ embryos appeared in November and larvae in February, _A. gelatinosum_ embryos in early February and larvae in March. The periods given above range from at least a month to five months. All the examples given breed in the colder months; development may be quicker in summer breeders.

Reproductive Seasons and Geographical Distribution

Hyman (1959 p.343) states "available data indicates that ectoprocts have an annual breeding season that extends over 2 or 3 up to 5 or 6 months. In the Northern Hemisphere the breeding season falls within the period from spring to autumn or even extends into early winter". Gautier (1962) has shown that this is not true of Mediterranean species and the data on reproduction given in Section II shows that Hyman's statement does not apply to the Manx area either. Many ectoproct species reproduce throughout the year; other species either reproduce only in the winter months or have their period of peak reproduction in the colder months (See Table 44). More species do reach peak reproductive activity in the warmer months but there is no scarcity of species bearing embryos in the colder months.

There is a little scattered data on the seasons of reproduction in other localities but these records usually apply to a few specimens and do not extend over the full breeding season of the species. Almost all the records of reproducing ectoprocts I have found in the literature fall within the season of reproduction of the species in Manx waters. The only data full enough for comparisons to be made with the Manx area is that given by Gautier (1962) for Mediterranean species. He gives data on the presence of embryos or eggs for 34 species which occur in Manx waters but in the majority of cases his records fall within the range of the species season of reproduction in the Isle of Man. *Bugula flabellata*, a species which breeds from May to October in the I.o.M., is recorded as reproducing in April and December in the Mediterranean. Gautier (1962) compares the reproduction of Mediterranean ectoprocts with those of the Channel and concludes that the season favourable for ectoproct reproduction is shorter in the Channel than in the Mediterranean. The sources from which he draws the data on reproduction of ectoprocts in the Channel are not complete. Table 45 shows that Manx ectoproct species reproduce throughout the year and that there is no break in ectoproct reproduction during the winter.

Of more interest is the relation between season of reproduction and geographical distribution. The data on this topic are summarized in Tables 46 & 47 from which several points emerge. 62% of widely-distributed species (i.e. species recorded from the Arctic and either or both the Mediterranean and the Tropics), and 68% of the species found in either or both the Mediterranean or Tropics but not the Arctic have embryos present throughout the year, but all the species recorded only from the Arctic and not the Mediterranean and Tropics have short breeding seasons and reproduce in the colder months. Species from warm water but not the Arctic tend to reach peak reproductive activity in Manx waters during the summer months when the water is at its warmest; widespread species do not appear to favour any particular season, some species reaching peak activity during each season. The data of Table 46 are of limited usefulness because of ~~its~~ their incomplete nature but, as far as ~~it~~ they go, ~~it~~ they appear to support that of Table 47. Thus the season of reproduction tends to vary with the geographical distribution of the species; species distributed in cold waters breed in winter; species from warm waters breed in summer.

The ability to reproduce at a wide range of temperatures is necessary if a species is to have a wide geographical distribution and it is noteworthy that many widely distributed species breed throughout the year in Manx waters.

Larval Colour and Taxonomy

Silén (1943) and Ryland (1958) have pointed out the usefulness of larval colour as a taxonomic character and in the present investigation notes have been made of larval colour wherever possible. The colours of embryos of a species have in all cases been constant although in some species there are slight differences in shade and intensity at different stages of development. Early embryos are often brighter in colour than late embryos. Late embryos may appear to have a bluish tinge but this is a structural colour associated with the cilia of the almost fully developed larva. Colours observed range from white, pale yellow and orange to red and maroon. No green or blue pigments have been seen. Embryo colour is a very useful character when working with fresh material; ~~but~~ the colour is lost on prolonged storage in alcohol but ~~however~~ it remains in weak formalin solution. The embryo colour is of use in many cases e.g. Escharella immersa has pink embryos, E. variolosa orange and E. ventricosa white; Cribrilina punctata pink and Cribrilina annulata yellow; Osthimosia avicularis yellow, Cellopora pumicosa red, etc. A full list of the embryo colours recorded is given in Appendix III.

Conclusions

1. The number of larvae produced by a species is correlated with the amount of protection given to the embryo and the length of planktonic larval life.

2. Species exposed to conditions on the shore where desiccation is likely tend to brood their embryos internally.

3. Shore species have short reproductive seasons.

4. Reproducing ectoprocts can be found at all times of the year. Arctic species reproduce during the colder months; warm-water species tend to breed during the warmer months but a proportion of warm-water species and widely distributed species reproduce throughout the year.

5. Larval colour is a useful taxonomic character.

SECTION V : RELATIONSHIPS BETWEEN ECTOPROCTS AND OTHER ANIMALS IN THE MANX AREA

I Associations

Many associations of varying degrees of constancy have been observed. Associations can be divided into two groups, those beneficial to the ectoproct and those beneficial to the other animal involved.

(a) Associations beneficial to the ectoproct

Many ectoprocts depend on other animals for supports. Many species are more or less confined to mollusc shells; some grow on living moluscs, others only on dead shells. In some cases the association is fairly specific e.g. the majority of *Smittoidea reticulata* were found on living *Anomia* spp., *Amphiblestrum flemingi* is very common on live *Chlamys opercularis*. Other species e.g. *Cellepораria dichotoma*, *Buskia nitens*, are dependent on hydroids for support. The most specific associations noted in the present investigation were those between *Hypophorella expansa* and the tubes of the polychaetes *Chaetopterus variopedatus* and *Lanice conchilega* and between *Triticella koreni* and the crustaceans *Calocaris macandreae* and *Nephrops norvegicus*.

(b) Associations beneficial to other animals

Animals occasionally shelter on ectoprocts. Numbers of *Porcellana longicornis* frequently occur on large "*Lepralia*" *foliacea* colonies. Turbellarians and nematodes were frequently found in dead zooecia. The tubes of *Lanice conchilega* from offshore frequently have *Cellaria sinuosa* fragments incorporated in their walls. Bushy *Cellaria fistulosa* colonies shelter amphipods and small worms. Caprellid amphipods are abundant on the turfs of erect ectoprocts found about and below ELWST in some localities. Ectoprocts provide supports for large numbers of folliculinids, Foraminiferans and hydroids. Very small scallops have been found attached by byssal threads to erect ectoprocts. Ectoprocts are frequently used as camouflage by spider crabs.

II Competitors with ectoprocts

Animals competing with ectoprocts can be divided into two groups: those competing for food and those competing for space. Some compete with ectoprocts for both food and space.

Filter-feeders such as sponges, sedentary worms, sea-squirts, lamellibranchs, folliculinids and perhaps hydroids, barnacles and *Porcellana* spp. etc. feed on small food organisms and suspended particles in the water and some or all must compete to a greater or lesser extent with ectoprocts for their food. In addition some of these, particularly sponges, ascidians, encrusting worms and barnacles compete with ectoprocts for space.

In some localities (e.g. Poyll Breine shore) sea-squirts and sponges are very abundant and ectoprocts virtually absent and in others ectoprocts may predominate (Port Erin Breakwater). This is probably because sponges and ascidians can cope with silt in suspension better than ectoprocts and therefore predominate on sheltered shores. Sponges can grow over ectoproct colonies e.g. on *Chlamys opercularis* but ectoprocts rarely grow on sponges. Goodbody (1961) noted the inhibition of development of sessile organisms on "Tuffnol" panels placed near a mature sponge-anemone-ophiuran community.

Anomia spp. frequently grow over and kill ectoprocts. Barnacles and sea anemones may settle on and kill ectoproct colonies. Ectoproct colonies frequently compete for space on the same support, if two co-specific colonies meet they fuse into a single colony but if two different species meet one may grow over the other and kill it.

III Predators of ectoprocts

(a) Predators of larvae

There is little information on this topic in the literature. I have seen ectoproct larvae pass into the water chambers of sponges and lamellibranchs, none of the larvae reappeared; they must have either been eaten or have become entangled in mucus. On the Modiolus bed off the south of the I.o.M. both Modiolus modiolus and large sponges are abundant but there are few ectoprocts present. The numbers of ectoproct larvae may be reduced by the activities of the Modiolus and the sponges. Other large filter-feeders as well as hydroids probably take a toll of ectoproct larvae. A Membranipora membranacea cyphonaute was found in the stomach of a Gobius flavescens (Fabr.) (P. J. Miller, personal communication).

(b) Predators of adult ectoprocts

Osburn (1921) gave evidence that ectoprocts are eaten by ducks and several fish including dogfish and sharks but points out that there is little information on this subject. Miller (1961) found that many nudibranchs feed on ectoprocts. Hartnoll (1961) found that some spider crabs use ectoprocts as articles of food as well as camouflage. Quasim (1957) showed that part of the diet of Blennius pholis L. in the Menai Straits consists of ectoprocts. I have found Crisidia cornuta and Crisia eburnea in stomachs of Blennius pholis. I found no ectoprocts in the stomachs of 12 Labrus bergylta Ascanius examined. Pycnogonids eat ectoprocts and are common on colonies of Cellaria fistulosa and Flustra foliacea and I have observed many nudibranchs feeding on ectoprocts. An arachnid is common on Flustrellidra hispida and Alcyonidium hirsutum colonies which frequently show damage apparently caused by the mite.

Conclusion

1. Ectoprocts form associations with other animals out of their need for support.

2. Some animals use ectoprocts as shelter.

3. Ectoprocts are eaten by various animals and compete with others for shelter and support.

SUMMARY OF RESULTS

1. 142 Ectoproct species have now been recorded from the Isle of Man. Several species new to Manx waters and two, *Alcyonidium variegatum* and *Penetrantia concharum*, new to British waters were collected in the present investigation.

2. The most important factors in the distribution of ectoprocts are availability of support and water movement.

3. Many ectoproct species are most abundant in areas of abundant hard support and considerable water movement.

4. Some species occur only in particular conditions and are 'indicators' of those conditions.

5. Ectoprocts are not distributed randomly, each species occurs on a more or less limited range of supports. Surface texture and area of the support are important.

6. The Manx ectoproct fauna contains more Mediterranean-Boreal species than Arctic-Boreal species.

7. Many species bear embryos throughout the year but Arctic species tend to reproduce in winter and southern species in summer.

8. Species which brood embryos internally are proportionately more abundant on the shore than elsewhere.

9. Ectoprocts compete with other animals for food and space and are preyed upon by various animals.

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Table 1-47

Table 1. *Crisiidae*. No. of colonies on the outer surface of dead shells for each 100 colonies on inner shell surfaces.

Species	No. of colonies on inner surface	No. of colonies on outer surface
Crisidia cornuta	100	49
Crisia aculeata	100	44
Crisia eburnea	100	101
Crisia ramosa	100	80
Crisia denticulata	100	109

Table 2. Crisiidae. Percentage distribution on supports from dredge samples.

SUPPORT	SPECIES: Crisidia cornuta	Crisia aculeata	Crisia eburnea	Crisia ramosa	Crisia denticulata
Chlamys	17·1	31·2	3·6	26·1	16·9
Pecten	16·2	22·5	·4	12·9	6·3
Cyprina	-	1·6	·3	4·4	2·1
Modiolus	2·7	5·6	·2	4·2	4·2
Glycymeris	19·8	15·6	·3	19·9	19·1
Other shell	6 3	5 4	2	8 6	-
Stone	4·5	7·9	·7	15·1	17·0
Hydroid	11·7	5·9	92·0	5·7	8·5
Cellaria	5·4	1·0	·6	1·2	14·9
Flustra	-	·3	·9	·3	-
Eucratea	4·5	-	·9	·8	8·5
Other supports	·6	3·5	·2	-	2·1
Total on shell	61·2	81·1	5·0	76·1	48·6
Total on zoophytes	33·3	7·2	94·0	8·0	31·9
Total	99·6	99·7	99·9	99·2	99·6

Table 3 .*Tubulipora penicillata*. Percentage distribution on supports dredged in areas A, B and C. Total support area examined: 97575 cm . No. of *T.penicillata* colonies: 105.

Support	% Composition of samples	% Distribution of *T.penicillata*
Chlamys	45·7	36·0
Modiolus	22·1	18.0
Pecten	9·8	16·1
Cyprina	6·1	9·5
Glycymeris	4·1	7·6
Gari	3·4	4·8
Stone	3·2	1·0
Cardium	1·3	-
Ensis	1·1	-
Other supports	2·6	7·0

Table 4 . *Diaperoecia major* and *Diaperoecia johnstoni*. Percentage distribution on supports from dredge samples. Total support surface area examined:575,000 cm . No. of *D.major* colonies: 387. No. of *D.johnstoni* colonies:527

Support	% Composition of samples	% Distribution of *D.major*	% Distribution of *D.johnstoni*
Chlamys	24·6	35·4	39·0
Pecten	24·3	26·8	26·7
Glycymeris	11·6	16·3	12·7
Stone	10·5	7·4	8·5
Modiolus	10·4	6·8	5·7
Cyprina	10·4	4·3	2·0
Buccinum	1·4	·5	·7
Lutraria	1·2	·3	-
Gari	1·0	·8	·6
Other supports	3·7	2·0	3·8

% *D.major* on inner surface of dead shell:	66·6
% *D.johnstoni* on inner surface of dead shell:	59·9
% *D.major* on outer surface of dead shell:	24·8
% *D.johnstoni* on outer surface of dead shell:	28·2
% *D.major* on living molluscs:	1·2
% *D.johnstoni* on living molluscs:	1·5

Table 5 . *Diplosolen obelia*, *Diastopora patina* and *Diastopora suborbicularis*. Percentage distribution on supports (excluding zoophytes) from dredge samples. Total support surface area examined: 575,000 cm . No. of *D.obelia* colonies:457. No. of *D.patina* colonies:607. No. of *D.suborbicularis* colonies: 5268

Support	% Composition of samples	% Distribution of *D.obelia*	*D.patina*	*D.suborbicularis*
Chlamys	24·6	6 3	40·0	21·5
Pecten	24·3	10·6	23·6	14·6
Glycymeris	11·6	9·6	6·7	12·9
Stone	11·3	8·0	·5	3·9
Modiolus	10·5	61·1	15·7	27·6
Cyprina	10·4	·2	2·1	7·8
Buccinum	1·4	·9	1·0	·2
Lutraria	1·2	-	·4	·9
Gari	1·0	·2	·4	4·4
Other supports	3·7	2·5	9·6	6·6

% *D.obelia* colonies on the inner surface of dead shells:* 32·4
% *D.patina* colonies on the ~~inium~~ surface of dead shells:* 58·4
% *D.suborbicularis* colonies inner surface of dead shells:* 54·9

% *D.obelia* colonies on the outer surface of dead shells:* 18·4
% *D.patina* colonies on the outer surface of dead shells:* 18·7
% *D.suborbicularis* on outer surface of dead shells:* 36·1

% *D.obelia* colonies on living molluscs: 40·3
% *D.patina* colonies on living molluscs: 21·0
% *D.suborbicularis* on living molluscs: 4·9

* lamellibranchs only

Table 6 *Diastopora suborbicularis*. Number of colonies on the outer surface of dead shell for each 100 colonies on the inner surface of each species of shell.

Shells are listed in order of decreasing roughness of the outer surface

Shell species	No. of colonies on inner surface	No. of colonies on outer surface.
Chlamys opercularis	100	33
Pecten maximus	100	34
Gari spp.	100	97
Cyprina islandica	100	145
Modiolus modiolus	100	113
Lutraria lutraria	100	78
Cardium crassum	100	100
Ensis spp.	100	103

Table 7 . *Lichenopora hispida*. Number of colonies on the outer surface of dead shell for each 100 colonies on the inner surface of each species of shell.

Shells are listed in order of decreasing roughness of the outer surface.

Shell species	No. of colonies on inner surface	No. of colonies on outer surface
Chlamys opercularis	100	22
Pecten maximus	100	45
Venus casina	100	38
Ostrea edulis	100	17
Gari spp.	100	54
Cyprina islandica	100	85
Modiolus modiolus	100	70
Lutraria lutraria	100	25
Cardium crassus	100	200
Ensis spp.	100	88

Table 8 . *Lichenopora hispida*. Percentage distribution on supports (excluding zoophytes) from dredge samples. Total surface area of support examined: 362,000 cm . No. of *L.hispida* colonies: 3575.

Support	% Composition of samples	% Distribution of *L.hispida*
Chlamys	26·1	31·1
Pecten	21·6	15·3
Stone	12·5	10·9
Modiolus	11·7	15·0
Glycymeris	10·8	16·0
Cyprina	7·9	2·1
Buccinum	1·7	1·7
Lutraria	1·7	·4
Gari	1·3	1·6
Dosinia	1·3	·3
Other supports	3·7	6·9

% *L.hispida* colonies on inner surface of dead shell:* 57·3
% *L.hispida* colonies on outer surface of dead shell:* 21·6
% *L.hispida* colonies on living molluscs: 8·3

* Lamellibranchs only.

Table 9. *Aetea anguinea* and *Aetea sica*. Distribution on supports in dredge samples. No. *A. anguinea* colonies: 97, No. *A. sica*: 846

Support	% Distribution *A. anguinea*	% Distribution *A. sica*
Hydroid	49·4	2·0
Cellaria	10·3	-
Eucratea	11·3	·6
Flustra	-	·1
Red algae	3·1	-
Stone	5·1	2·7
Inner shell surface	9·3	22·8
Outer shell surface	11·3	63·0
Live molluscs	-	8·4

Table 10 . *Aetea sica*. Number of colonies on the outer surface of dead shell for each 100 colonies on the inner surface of each species of dead shell.

Shells are listed in order of decreasing roughness of the outer surface.

Shell species	No. of colonies on inner surface	No. of colonies on outer surface
Chlamys opercularis	100	327
Pecten maximus	100	138
Gari spp	100	166
Cyprina islandica	100	800
Modiolus modiolus	100	428

Table 11 . *Aetea sica*. Percentage distribution on supports from dredge samples. Total support surface area examined: 362,000 cm . Number of *A.sica* colonies: 846.

Support	% Composition of samples	% Distribution of *A.sica*
Chlamys	26·1	47·9
Pecten	21·6	10·6
Stone	12·5	2·7
Modiolus	11·7	15·9
Glycymeris	10·8	8·4
Cyprina	7·9	4·4
Buccinum	1·7	·5
Lutraria	1·7	1·8
Gari	1·3	2·9
Dosinia	1·0	-
Other supports	3·7	1·3

excluding zoophytes

Table 12 . *Eucratea loricata*. Percentage distribution on supports from dredge samples. No. of *E.loricata* examined: 100

Support	% colonies of *E.loricata*.
Chlamys opercularis	49
Glycymeris glycymeris	14
Pecten maximus	11
Modiolus modiolus	3
Cyprina islandica	2
Venus fasciata	1
Fragments	1
Zoophytes	19

% colonies on inner shell surface: 5
% colonies on outer shell surface: 72
% colonies on living molluscs : 4

Table. 13 . The influence of the hermit crab *Pagurus bernhardus* on the frequency of occurence of the ectoproct species commonly occurring on the gastropods *Buccinum undatum*, *Neptunea antiqua* and *Colus gracilis*.

Ectoproct species	Live gastropod *Buccinum* (129)	Live gastropod *Neptunea* (54)	Broken, empty shells *Buccinum* (17)	Broken, empty shells *Neptunea* (2)	Empty shells *Buccinum* (96)	Empty shells *Neptunea* (13)	Empty shells *Colus* (2)	Shells with *P. bernhardus* *Buccinum* (139)	Shells with *P. bernhardus* *Neptunea* (8)	Shells with *P. bernhardus* *Colus* (8)
	Description of shell, number examined and % occurrence of ectoproct species									
Electra pilosa	7	-	6	-	24	-	50	53	50	50
Scrupocellaria reptans	-	-	29	50	-	18	-	-	-	-
Escharella immersa	27	7	47	-	50	18	-	26	25	25
Schizomavella auriculata	19	24	35	50	34	18	-	12	12	-
Microporella ciliata	5	-	53	-	30	-	-	28	-	-
Porella concinna	15	11	35	-	19	18	-	14	25	-
Osthimosia avicularis	6	2	35	-	13	6	-	8	12	-
Parasmittina trispinosa	3	1[illegible]	18	-	13	-	-	4	-	-
Alcyonidium mytili?	5	-	-	-	6	-	-	14	-	-

Table 14 . *Pyripora catenularia*. Percentage distribution on supports from dredge samples.

Support	% Distribution of *P.catenularia*.
Inner surface of dead shell	34·5
Outer surface of dead shell	26·9
Live molluscs	2.0
Shell fragments	11·3
Stone	25·1
Hydroid	·2

Table 15 . *Pyripora catenularia*, *Alderina imbellis*, *Amphiblestrum flemingi* and *Callopora lineata* . Number of colonies of each species on the outer surface of dead shell for each 100 colonies on the inner surface of each species of dead shell.

Shell species are listed in order of decreasing roughness of outer shell surface.

Shell species	Number of colonies on inner surface	Number of colonies on outer surface *P.catenularia*	*A.imbellis*	*A.flemingi*	*C.lineata.*
Chlamys opercularis	100	53	18	159	14
Pecten maximus	100	22	20	22	10
Glycymeris glycymeris	100	4	-	14	4
Cyprina islandica	100	-	100	-	66
Modiolus modiolus	100	82	100	57	55
Lutraria lutraria	100	-	-	14	-

Table 16 . *Alderina imbellis* and *Amphiblestrum flemingi*. Percentage distribution on supports from dredge samples. Total area of supports examined 362,000 cm . No. of *A. imbellis* colonies 673 No. of *A. flemingi* colonies 508.

Support	% Composition of samples	% Distribution of *A. imbellis*	% Distribution of *A. flemingi*.
Chlamys	26·1	45·0	52·8
Pecten	21·6	26·8	5·9
Stone	12·5	3·0	2·7
Modiolus	11·7	8·9	16·1
Glycymeris	10·8	·9	10·4
Cyprina	7·9	11·4	-
Buccinum	1·7	·3	·3
Lutraria	1·7	·6	-
Gari	1·3	·2	·1
Dosinia	1·0	·4	-
Cardium crassum	·7	1·0	-
Ensis	·5	-	-
Other supports	2·5	1·9	11·6

% *A. imbellis* on inner shell surfaces; 71·8
% *A. flemingi* on inner shell surfaces: 33·0

% *A. imbellis* on outer shell surfaces: 23·2
% *A. flemingi* on outer shell surfaces: 37·6

% *A. imbellis* on living molluscs: 1·9
% *A. flemingi* on living molluscs: 26·2

Table 17 *Scrupocellaria scruposa*. Number of colonies on the outer surface of dead shell for each 100 colonies on the inner surface of each species of dead shell.

Shell species are listed in order of decreasing roughness of the outer surface.

Species of shell	No. of colonies on inner surface	No. of colonies on outer surface.
Chlamys opercularis	100	292
Pecten maximus	100	156
Glycymeris glycymeris	100	44
Cyprina islandica	100	750
Modiolus modiolus	100	383

Table 18 *Scrupocellaria scruposa*. Distribution on supports from areas A,B and C. Surface area of supports examined 97575 cm , number of *S. scruposa* colonies; 358.

SUPPORT	% COMPOSITION OF SUPPORTS	% DISTRIBUTION OF *S. SCRUPOSA*
Chlamys	41·3	36·3
Modiolus	22·2	14·8
Pecten	9·9	10·0
Cyprina	6·2	2·7
Live *Chlamys*	4·7	8·9
Glycymeris	4·2	13·1
Gari	3·4	·6
Stone	3·2	1·9
Cardium crassum	1.3	·8
Ensis	1.1	-
Buccinum	·6	-
Lutraria	·6	1·1
Zoophytes	?	5·0
Other supports	1·3	4·8

% *S. scruposa* on inner shell surfaces: 266
% *S. scruposa* on outer shell surfaces 56·4
% *S. scruposa* on living mollusca 8·9

Table 19. Percentage distribution of *Bicellariella ciliata* on dredged supports. No. of *Bicellariella ciliata* colonies examined: 249

Support	% Distribution of *B.ciliata*
Inner surface of dead shell	8·8
Outer surface of dead shell	38·2
Living molluscs	7·3
Stone	1·2
Zoophytes	43·9
Other supports	·8

Table 20 . Hippothoa distans and Chorizopora brongniarti. Number of colonies on the outer surface of dead shell foreach 100 colonies on the inner surface of each species of dead shell.
Shell species are listed in order of decreasing rougness of the outer surface.

Shell species	No. of colonies on inner surface	No. of colonies on outer surface H.distans	C.brongniarti
Chlamys opercularis	100	12	18
Pecten maximus	100	13	16
Gari spp	100	56	18
Glycymeris glycymeris	100	12	47
Cyprina islandica	100	56	220
Modiolus modiolus	100	64	65
Ensis spp.	100	-	45

Table 21 . *Hippothoa distans* and *Chorizopora brongniarti*
Pecentage distribution on supports from dredge samples. Total support surface area examined; 575,000 cm . No. of *H.distans* colonies: 1,429. No. of *C. brongniarti* colonies:990.

Support	% Composition of samples	% Distribution of *H.distans*	% Distribution of *C.brongniarti*
Chlamys	24·6	38·8	33·3
Pecten	24·3	22·2	19·4
Glycymeris	11.6	18·2	23·9
Stone	11.3	2·6	1·7
Modiolus	10·5	11·2	8.5
Cyprina	10.4	2·3	5·2
Buccinum	1·4	·3	·2
Lutraria	1·2	·8	·7
Gari	1.0	1·6	1·3
Dosinia	·7	-	·1
Clinker	·5	-	-
Ensis	·3	·2	1·6
Other shell	2·2	9·9	4·4

% *H.distans* on inner surface of shell: 77 5
% *C. brongniarti* on inner shell surface : 76 7

% *H.distans* on outer shell surface : 15 5
% *C.brongniarti* on outer shell surface: 18 0

% *H. distans* on living molluscs : 4 0
% *C.brongniarti* on living molluscs: 3 3

Table 22 *Reptadeonella violacea.* Percentage distribution on supports from dredge samples. Total support surface area examined : 575,000 cm . No. of *R.violacea* colonies: 190.

Support	% Composition of samples	% Distribution of *R.violacea*.
Chlamys	24.6	24.7
Pecten	24.3	4.2
Glycymeris	11.6	33.1
Stone	11.3	28.4
Modiolus	10.5	3.1
Cyprina	10.4	.8
Buccinum	1.4	-
Lutraria	1.2	1.6
Gari	1.0	.8
Dosinia	.7	-
Clinker	.5	-
Ensis	.3	-
Venus casina	.3	1.6
Chlamys distorta	.2	-
Other supports	.7	-

% *R.violacea* on inner shell* surfaces: 51.5
% *R.violacea* on outer shell* surfaces: 20.2
% *R.violacea* on living molluscs -

*lamellibranchs only.

Table 23. *Escharella immersa*, *Escharella ventricosa*, and *Escharella variolosa*. Number of colonies on the outer surface of dead shells for each 100 colonies on the inner surface of each type of shell.

Shell species are listed in order of decreasing roughness of the outer surface.

Species of shell	No. of colonies on inner surface	No. of colonies on outer surface *E. immersa*	*E. ventricosa*	*E. variolosa*
Chlamys opercularis	100	56	85	47
Pecten maximus	100	42	38	9
Venus casina	100	84	-	-
Gari spp.	100	77	11	-
Glycymeris glycymeris	100	44	18	7
Cyprina islandica	100	50	176	-
Modiolus modiolus	100	-	55	50
Lutraria lutraria	100	68	28	-
Cardium crassus	100	-	6	-
Ensis spp.	100	35	-	-

Table 24 . *Escharella immersa* . Percentage distribution on supports from dredge samples. Total support surface area examined: 362,000 cm . No. of *E. immersa* colonies: 9275

Support	% Composition of samples	% Distribution of *E.immersa*.
Chlamys	26·1	15·8
Pecten	21·6	6·4
Stone	12·5	29·7
Modiolus	11·7	18·4
Glycymeris	10·8	22·1
Cyprina	7·9	1·1
Buccinum	1·7	·8
Lutraria	1·7	·4
Gari	1·3	·5
Dosinia	1·0	·1
Cardium crassum	·7	·3
Ensis	·5	·3
Venus casina	·5	·5
Ostrea	·5	·1
Other shells	1·5	3·8

% *E.immersa* on inner shell surfaces: 40·0

% *E.immersa* on outer shell surfaces: 21·4

% *E.immersa* on living molluscs : 7·5

Table 25 : *Escharella ventricosa* and *Escharella variolosa*. Percentage distribution on supports from dredge samples. Total surface area examined: 575,000 cm . No. of *E.ventricosa* colonies: 2393. No. of *E.variolosa* colonies: 319

Support	% Composition of supports	% Distribution of *E.ventricosa*	% Distribution of *E.variolosa*.
Chlamys	24·3	38·8	18·6
Pecten	24·3	25·4	7·2
Glycymeris	11·6	7·2	37·8
Stone	11·3	11·7	10·3
Modiolus	10·5	6·2	15·7
Cyprina	10·4	4·8	·6
Buccinum	1·4	·4	1·2
Lutraria	1·2	·7	·6
Gari	1·0	·8	-
Dosinia	·7	·1	-
Clinker	·5	·4	·6
Ensis	·3	·2	-
Venus casina	·3	·4	-
Chlamys distorta	·2	·1	1·3
Other shell	1·7	2·8	5·8

% *E.ventricosa* on inner shell surfaces 68·9
% *E.variolosa* on inner shell surfaces 61·7

% *E.ventricosa* on outer shell surfaces 17·5
% *E.variolosa* on outer shell surfaces 12·8

% *E.ventricosa* on living molluscs 1·5
% *E.variolosa* on living molluscs 9·4

Table 26. Schizoporella unicornis, Schizomavella auriculata and Schizomavella linearis. Percentage distribution on supports from dredge samples. Total area of supports examined : 575,000 cm . No. of S. unicornis colonies: 232. No. of S. auriculata colonies: 6905. No. of S. linearis colonies: 799

Support	% Composition of samples	% Distribution of S. unicornis	S. auriculata	S. linearis.
Chlamys	24·6	13·7	35·0	24·9
Pecten	24·3	7·3	23·8	13·9
Glycymeris	11·6	41·5	10·8	23·2
Stone	11·3	26·8	4·8	6·6
Modiolus	10·5	5·2	9·5	21·4
Cyprina	10·4	6	7·1	·1
Buccinum	1·4	2·1	·7	·7
Lutraria	1·2	·4	·9	·4
Gari	1·0		1·2	·4
Dosinia	·7		2	
Clinker	·5		·1	·5
Ensis	·3		·5	
Other supports	2·2	3·0	5·3	7·9

% S. unicornis on inner surface of dead lamellibranch shells : 61·2
% S. auriculata on inner surface of dead lamellibranch shells : 48·2
% S. linearis on inner surface of dead lamellibranch shells : 55·4

% S. unicornis on outer surface of dead lamellibranch shells : 9·5
% S. auriculata on outer surface of dead lamellibranch shells : 44·0
% S. linearis on outer surface of dead lamellibranch shells : 26·4

% S. unicornis on live molluscs : –
% S. auriculata on live molluscs 1·5
% S. linearis on live molluscs 10·0

Table 27 . Schizomavella auriculata and Schizomavella linearis. Number of colonies on the outer surface of dead shells for each 100 colonies on the inner surface.

Shell species are listed in order of decreasing roughness of the outer surface of their shells.

Species of shell	No. of colonies on inner surface	No. of S. auriculata colonies on the outer surface	No. of S. linearis colonies on the outer surface
Chlamys opercularis	100	90	56
Pecten maximus	100	73	29
Venus casina	100	118	
Ostrea edulis	100	100	
Dosinia lapillus	100	100	
Gari spp.	100	97	
Glycymeris glycymeris	100	38	49
Cyrrina islandica	100	91	
Modiolus modiolus	100	179	45
Lutraria lutraria	100	134	
Cardium crassus	100	52	
Ensis spp.	100	13	

Table 28 . *Hippoporina pertusa.*
Percentage distribution on supports from dredge samples. Total surface area of supports examined: 575,000 cm . No. of *H.pertusa* colonies: 438

Support	% Composition of samples	% Distribution of *H.pertusa*
Chlamys	24·6	50·1
Pecten	24·3	27·9
Glycymeris	11·6	·4
Stone	11·3	1·8
Modiolus	10·5	1·4
Cyprina	10·4	17·4
Buccinum	1·4	·9
Lutraria	1·2	·2
Gori	1·0	.6
Dosinia	·7	-
Clinker	·5	-
Ensis	·3	1·0
Other supports	2·2	2·3

% *H.pertusa* colonies on inner surface of dead lamellibranchs: 43·1
% *H.pertusa* colonies on outer surface of dead lamellibranchs : 51·8
% *H.pertusa* colònies on living molluscs : 2·4

Table 29 . Microporella ciliata and Fenestrulina malusi. Distribution on supports from dredge samples. Total support surface area examined: 575,000 cm . No. of M.ciliata colonies: 8396. No. of F.malusi colonies: 7109

Support	% Composition of samples	% Distribution of M.ciliata	% Distribution of F.malusi
Chlamys	24·6	24·8	32·4
Pecten	24·3	12·0	14·9
Glycymeris	11·6	31·4	28·6
Stone	11·3	13·5	4·4
Modiolus	10·5	11·0	8·6
Cyprina	10·4	1·2	1·4
Buccinum	1·4	·9	·4
Lutraria	1·2	1·0	1·6
Gari	1·0	·9	1·3
Dosinia	·7	·1	·2
Clinker	·5	–	–
Ensis	·3	·4	·6
Other supports	1·7	2·2	4·6

% *M.ciliata* on inner surface of dead lamellibranch shells: 60·0
% *F.malusi* on inner surface of dead lamellibranch shells: 83·4

% *M.ciliata* on outer surface of dead lamellibranch shells: 21·8
% *F.malusi* on outer surface of dead lamellibranch shells: 8·3

% *M.ciliata* on living molluscs: 3·3
% *F.malusi* on living molluscs: 2·8

Table 3c . *Microporella ciliata* and *Fenestrulina malusi.*
Number of colonies on the outer surface of dead shells for each 100 colonies on the inner surface of each type of shell.
Shell species are listed in order of decreasing roughness of the outer surface of the shell .

Species of shell.	No. of colonies on inner surface	No. of *M.ciliata* colonies on outer surface	No. of *F.malusi* colonies on outer surface.
Chlamys opercularis	100	25	10
Pecten maximus	100	24	21
Venus casina	100	80	15
Dosinia lapillus	100		25
Ostrea edulis	100	-	-
Gari spp.	100	40	12
Glycymeris glycymeris	1000	33	5
Modiolus modiolus	100	79	22
Lutraria lutraria	100	94	14
Cardium crassus	100	50	-
Ensis spp.	100	67	57
Add. *Cyprina islandica*	100	179	4

Table 31. *Porella concinna* and *Parasmittina trispinosa*. Distribution on supports from dredge samples. Total surface area of supports examined: 575,000 cm. No. of *P. concinna* colonies: 5098. No. of *P. trispinosa* colonies: 1658

Support	% Composition of supports	Percentage distribution of *P. concinna*	Percentage distribution of *P. trispinosa*
Chlamys	24.6	41.5	27.3
Pecten	24.3	20.0	18.2
Glycymeris	11.6	11.0	30.3
Stone	11.3	6.3	9.2
Modiolus	10.5	8.9	7.9
Cyprina	10.4	4.9	1.4
Buccinum	1.4	.5	.3
Lutraria	1.2	.7	.5
Gari	1.0	1.3	1.0
Dosinia	.7	.1	.4
Clinker	.5	.2	.1
Ensis	.3	.2	-
Venus casina	.3	.4	.2
Chlamys distorta	.2	.1	.1
Other shell	1.7	3.8	3.1

% *P. trispinosa* on inner shell* surface	51.7
% *P. concinna* on inner shell* surface	38.9
% *P. trispinosa* on outer shell* surface	35.0
% *P. concinna* on outer shell* surface	51.2
% *P. trispinosa* on living shells	.5
% *P. concinna* on living shells	2.4
% *P. trispinosa* on other shells	.5
% *P. concinna* on other shells	.9

*lamellibranchs only.

Table 32 . *Porella concinna* and *Parasmittina trispinosa*. No. of colonies on the outer surface of dead shells for each 100 colonies on the inner shell surface.

Species of shell	No. of colonies on inner surface	No. of colonies *P. concinna* on outer surface.	No. of colonies *P. trispinosa* on outer surface.
Chlamys opercularis	100	99	76
Pecten maximus	100	214	65
Venus casina	100	170	-
Ostrea edulis	100	88	-
Gari spp.	100	144	31
Glycymeris glycymeris	100	92	54
Cyprina islandica	100	280	156
Modiolus modiolus	100	200	115
Lutraria lutraria	100	136	-
Cardium crassus	100	110	-
Ensis spp.	100	100	-

Shells species decrease in roughness of outer shell surface from top of table towards bottom.

Table 33 . _Celleporaria pumicosa_ and _Osthimosia avicularis_. Number of colonies on the outer surface of dead shells for each 100 colonies on the inner surface of the shells. Shell species decrease in roughness of the outer shell surface from the top of the table towards the bottom.

Species of shell	No. of colonies on inner surface	No. of colonies _C. pumicosa_ on outer surface	No. of colonies _O. avicularis_ on outer surface
Chlamys opercularis	100	175	310
Pecten maximus	100	56	111
Gari spp.	100	34	25
Glycymeris glycymeris	100	54	64
Cyprina islandica	100	100	275
Modiolus modiolus	100	450	155

Table 34 Cellepornria pumicosa and Osthimosia avicularis. Percentage distribution on supports other than zoophytes from dredge samples. Total surface area of supports examined : 575,000 cm . No. of C.pumicosa colonies: 372. No. of O. avicularis: 1230.

Support	% composition of supports.	% distribution of C.pumicosa	% distribution of O.avicularis
Chlamys	24·6	38·9	63·4
Pecten	24·3	6·8	7·4
Glycymeris	11·6	14·6	11·8
Stone	11·3	7·9	4·1
Modiolus	10·5	12·7	7.0
Cyprina	10·4	3·8	1·2
Buccinum	1·4	-	·9
Lutraria	1·2	4·5	·1
Gari	1·0	[illegible]	·8
Dosinia	·7	1·1	-
Clinker	·5	-	-
Ensis	·3	1.1	·3
Other support	2·2	8·6	2 8

% C.pumicosa on inner shell* surfaces : 33·6
% O. avicularis on inner shell* surface: 27·4

% C.pumicosa on outer shell* surfaces : 45·4
% O.avicularis on outer shell* surfaces: 54·6

% C. pumicosa on living shells : 9.1
% O avicularis on living shells : 2·2

* lamellibranchs only

Species	Support: Algae	Zoophytes	Shells	Stones	Crustacea	Bottom type: Shore	Rocky sub-littoral	Coarse gravel	Sandy shelly gravel	Inshore muddy sand	Offshore muddy sand	Mud
Eucratea loricata		+	+++				?	+++	+	+	+	
Scrupocellaria reptans	+++	+	++	+++		++	+++	+	+	+	+	
S.scrupea			+++					+++	+	+	+	
S.scruposa		+	+++	+			?	+++	++	+	+	
Bicellariella ciliata		+++	++			+		+++	++	++	+	
Bugula avicularia		++	+++	+			?	+++	++	++		
B.plumosa		+++	+++	+				+++	++		+	
B.flabellata.		+++	+++	++			?	+++	+++	++	+	
Flustra foliacea			+++	++			?	+++	+			
*Cellaria fistulosa		+++	+++	+			?	+++	++	++	++	
*C.sinuosa		+++	+++	+			?	+++	++	++	++	
Lepralia foliacea			+	+++				+++				
Palmicellaria skenei			+++					+			+++	
Celleporaria dichotoma		+++						+	+	+	+++	
Crisidia cornuta	+++	++	+++	++	+	++	+++	++	+	+	+	+
Crisia eburnea	+++	+++	+	+		+	+++	+++	++	+	+	
C.aculeata		+	+++	+++	+	+	?	+++	++	+	++	+
C. denticulata		+	+++	+++		+	?	+++	++	+		
C.ramosa		++	+++	+++			?	+++	+++	+	+	
Tubulipora penicillata			+++	+				+++	+	+		
Alcyonidium gelatinosum			+++	+				++	+++	+	++	

+++ Common ++ Occasional + Rare
*on zoophytes for early part of life.

Table 35a, Distribution of erect ectoprocts by support and by bottom type. Ratings are assessed individually for each species and do not allow direct comparison of abundance between species. Each species is given a +++ rating for the support and bottom type on which it is most common.

Rare erect species not included in Table 35a are: Filicrisia geniculata, Entalophora clavata, Bugula calathus, Bugula turbinata, Cellaria salicornioides, Porella compressa, Omalosecosa ramulosa, Vesicularia spinosa, Amathia lendigera, Bowerbankia pustulosa.

	Support					Bottom Type						
Ectoproct species	Algae	Zoophytes	Shells	Stones	Crustacea	Shore	Rocky sub-littoral	Coarse gravel	Sandy shelly gravel	Inshore muddy sand	Offshore muddy sand	Mud
† Tubulipora liliacea		+++	+					+	+	+	+++	
† Tubulipora phalangea			+++	+++		+++	+++	++	+	+	+	
† Diaperoecia major			+++	+				+++	++		+++	
† Aetea anguinea	+	+++	+	+		+	+	+++	+	+	+	
† Aetea sica	+	+	+++	+		+	?	+++	+++	+	+	
† Aetea truncata	+	+++	+					+	++	+++		
† Scruparia chelata	++	+++	+			+++	?	+	+	+		
† Scruparia ambigua	++	+++	+			+++	?	+	+	+	+	
Pyripora catenularia			++	+++			?	+++	++	+		
† Beania mirabile	+	++	+++	+		+	+	+++	++	+	+	
Hippothoa distans			+++	+				+++	+	+	+	
Hippothoa divaricata			++	+++		+	+	+++	+	+	+	
† Alcyonidium mammillatum			++	+++		+	?	4++	++			
✕ Arachnidium hippothooides			+++					++			+++	
† Bowerbankia imbricata	+	+	+	+++		+++	?	+				
† Bowerbankia gracilis		+++		+		+++	?	+	+	+	+	
Buskia nitens		+++							+		+++	
† Valkeria uva	+++			+		+++						
† Valkeria tremula		+++	+						+ +	++	+	
† Triticella koreni					+++							+++
† Nolella dilatata		++	+++	+				+++	++	+	+	

+++ Common ++ Occasional + Rare

† species in which the tentacular crown is raised above support

✕ common on Ascidians

Rare species and species of unknown distribution not included:
Stomatopora granulata, S. incurvata, Diaperoecia johnstoni, Tubulipora lobulata, Proboscina incrassata, Nolella pusilla

Table 35b. Distribution of spreading ectoproct species by support and by bottom type. Assessments as for Table 35a.

	Support					Bottom Type						
	Algae	Zoophytes	Shells	Stones	Crustacea	Shore	Rocky sub-littoral	Coarse gravel	Sandy shelly gravel	Inshore muddy sand	Offshore muddy sand	Mud
Diplos												
Diplosolen obelia S			+++	++		+	?	+++	++			
Diastopora patinaS	+	++	+++	+		+	?	+++	+++	+	+	
D.suborbicularis S			+++	+++		+	++	+++	+++	++	++	
Lichenopora hispida S		+	+++	++	+	+	++	+++	+++	+++	++	
'Callopora aurita S				+++		+++	?					
C.craticula S	+		+++				?			+++		
C.dumerili S			+++	+		+	?	+++	+++	+	+	
C.lineata S	+++		+	++		++	+++	+	+	+		
Amphiblestrum flemingi S			+++	+		+	?	++	+++	+	+	
Alderina imbellis S			+++	+				+	++	+++	+++	
Membranipora membranacea	+++					+++	+++					
"Electra pilosa L?	+++	+++	++	++	++	+++	+++	++	++	++	++	++
Cauloramphus spiniferum S			+	+++		+++	?	+				
Cribrilina radiata L			+++	+			?	+++	++			
"C.punctataS			+++	+++		+++	?	+		+		
Figularia figularis L			+++					+++	++			
Membraniporella nitida S			+	+++		+++	?	++	+	+		
Microporella ciliata S.	+++	+	++	++		+++	++	+++	++	+	+	
Fenestrulina malusi S			+++	+				++	+++	++	+	
Reptadeonella violacea L			+	+++				+++	+			
Hippothoa hyalina S	+++	+		+	+	+++	+++	+	+	+		
Chorizopora brongniarti S			+++	+		+	?	+++	++	+	+	+
Haplopoma graniferum L				+++		+++	?					
Cryptosula pallasiana L	+			+++		+++	?					
Hippoporina pertusa S.			+++					+	++	+++	+++	
Umbonula littoralis S				+++		+++	?					
Porella concinnaL			+++	+			?	++	+++	++	++	
Smittina cheilostomata S			+++	+++				+++	++	+		
S.lan'sborovi L			+++	+				++	+++	+		
Smittoid a reticulata L			+++	+				+++	++	+		
Parasmittina trispinosa L			+++	++		+	++	++	+++	+	+	
Escharoides coccineus S	+		+	+++		+++	?	+	+			
Escharella immersa S.			+++	+++	+	+++	+++	+++	++	+	+	+
E.variolosa L			+++	+			?	+++	+++	+		
E.ventricosa S			+++	++				++	+++	++	+++	+
Shizoporella unicornis S			+	+++		+++	++	+	+			
Schizomavella auriculata S			+++	+	+			++	+++	+++	+++	+
S.linearis S			+++	++		+++	?	++	+	+	+	
Schizotheca fissa S			+++	+				+++	+		+	
Escharina spiniferum L	+		+	+++		+++	++	++				
Alcyonidium hirsutum S	+++					+++						
A.mytili S			++	+++		+++	?	+++	+			
A.variegatum S			+++	+				+	+++	++		
Flustrellidra hispida S	+++					+++			++			

+++ Common ++ Occasional + Rare
'some colonies on Laminaria holdfasts ˣ1 colony on Zoophyte
"1 colony on shore S colony can be mature at small size L colony large at maturity

Table 35c. Distribution of encrusting ectoprocts (excluding nodulous and spreading species). Ratings assessed as for Table 35a.

Species not included in Table 35c include: Tubulipora aperta, Lichenopora radiata, L.verrucaria, Conopeum reticulum, Callopora discreta, Rosseliana rosseli, Ammatophora nodulosa, Amphiblestrum solidum, Hincksina flustroides, Cribrilina annulata, Phylactolla collaris, P.labrosa, Umbonula arctica, Lagenipora lepraloides, Lepralia adpressa, Porella minuta, Schizoporella discoidea, Escharina johnstoni, E.vulgaris, E.alderi, Herentia hyndmanni etc.

Table 35d Distribution of nodular ectoprocts by support and by bottom type. Ratings assessed as for Table 35a.

	Osthimosia avicularis	*Celleporaria pumicosa*	*Celleporina costazii*
Support			
Algae			+
Zoophytes	+++	+	+++
Shells	++	++	+
Stones	+	+++	+++
Crustacea			
Bottom Type			
Shore		+++	+++
Rocky sub-littoral		?	?
Coarse gravel	++	++	+++
Sandy shelly gravel	+++	+++	++
Inshore muddy sand	+	++	+
Offshore muddy sand	+	+	+++
Mud			

+++ Common ++ Occasional + Rare

Rare species not included in Table 35d : *Hippoporidra edax*, *Osthimosia armata*

Table 36 . Ectoprocts growing on algae in Manx waters. Rare species are excluded.

Group I. Species occasionally growing on algae, most common on other supports.

Colony Type

Erect	Spreading	Encrusting	Nodular
-	*Aetea anguina*	*Callopora craticula*	*Cellaporina costazii*
	Aetea sica	*Escharina spiniferum*	
	Aetea truncata	*Escharoides coccineus*	
	Beania mirabilis		
	Bowerbankia imbricata		

Group II. Species regularily present on algae.

Colony Type

Erect	Spreading	Encrusting
Crisidia cornuta	*Scruparia ambigua*	*Callopora lineata*
Crisia eburnea	*Scruparia chelata*	*Membranipora membranacea*
Scrupocellaria reptans	*Valkeria uva*	*Electra pilosa*
		Microporella ciliata
		Hippothoa hyalina
		Alcyonidium hirsutum
		Flustrellidra hispida

Table 37 Ectoprocts growing on zoophytes in the Manx area.

*Group I . Species occasionally present on zoophytes but most common on other supports.

Colony Type

Erect	Spreading	Encrusting	Nodular
Crisia aculeata	Aetea sica	Lichenopora hispida	-
Crisia denticulata	Bowerbankia imbricata	Microporella ciliata	
Crisia ramosa		Hippothoa hyalina	
Eucratea loricata			
Scrupocellaria reptans			
Scrupocellaria scruposa			

*Group II Species growing on zoophytes but which are rare in the area sampled

Colony Type

Erect	Encrusting
Omalosecosa ramulosa	Alcyonidium parasiticum

Group III Species common on zoophytes in the area sampled and which usually grow on or close to the axis of erect zoophyte colonies.

Colony Type

Erect	Spreading	Encrusting	Nodular
Crisidia cornuta	Aetea anguina	Diastopora patina	Osthimosia avicularis
Crisia eburnea	Aetea truncata	Electra pilosa	
Bugula avicularia	Scruparia chelata		
Bugula plumosa	Scruparia ambigua		
Cellaria fistulosa	Beania mirabilis		
Cellaria sinuosa	Bowerbankia gracilis		
Cellaporaria dichotoma	Buskia nitens		
	Valkeria tremula		
	Nolella dilatata		

Group IV Species common in the area sampled and occurring most commonly on the lateral branches of zoophyte colonies;

Erect	Nodular
~~Bicellariella~~ Bicellariella ciliata	Celleporina costazii

Table 38 Ectoprocts colonizing shell in Manx waters (excluding rare species)

Group I Species rare on shell.

Erect	Spreading	Incrusting: Large	Incrusting: Small	Nodular
Lepralia foliacea	Tubulipora liliacea	Reptadeonella violacea	Cauloramphus spiniferum	Celleporina costazii
	Aetea anguina		Hippothoa hyalina	
	Scruparia spp.		Cribrilina punctata	
	Valkeria tremula		Schizoporella unicornis	
			Membraniporella nitida	

Group II. Species common on shell but also common on other supports.

Erect	Spreading	Incrusting: Large	Incrusting: Small	Nodular
Eucratea loricata	Tubulipora phalangea	Porella concinna	Diplosolen obelia	Osthimosia avicularis
Scrupocellaria scruposa	Aetea sica	Parasmittina trispinosa	Diastopora patina	Cellepora pumicosa
Scrupocellaria reptans	Pyripora catenularis	Escharella variolosa	Diastopora suborbicularis	
Bicellariella ciliata	Beania mirabilis	Electra pilosa	Lichenopora hispida	
Bugula flabellata	Hippothoa divaricata		Callopora craticula	
Flustra foliacea	Alcyonidium mamillatum		Callopora lineata	
Cellaria fistulosa	Arachnidium hippothooides		Microporella ciliata	
Cellaria sinuosa	Nolella dilatata		Escharella immersa	
Crisidia cornuta			Schizomavella auriculata	
Crisia eburnea			Schizomavella linearis	
Crisia aculeata			Escharoides coccineus	
Crisia denticulata			Alcyonidium mytili	

Group III. Species commonly present on shell, rare on other supports

Erect	Spreading	Incrusting: Large	Incrusting: Small
Palmicellaria skenei	Diaperoecia major	[illegible]laria [illegible]ularis	Fenestrulina malusi
Bugula avicularia	Hippothoa distans	Cribrilaria [illegible]	Alderina imbellis
Bugula plumosa		Smittina landsborovi	Amphiblestrum flemingi
Scrupocellaria scruposa		Smittoidea reticulata	Callopora dumerili
Tubulipora penicillata			Chorizopora brongniarti
Alcyonidium gelatinosum			Hippoporina pertusa
			Smittina cheilostomata
			Schizotheca fissa
			Alcyonidium variegatum

Table 39. Ectoprocts colonizing stone in Manx waters.(Shore records not included).

Group I. Species rarely occurring on stones.

Colony Type

Erect	Spreading	Incrusting: Large	Incrusting: Small	Nodular
Bugula avicularia	Diaperoecia major	Cribrilaria radiata	Alderina imbellis	Osthimosia avicularis
Bugula plumosa	Aetea anguina		Hippothoa hyalina	
Crisia eburnea	Hippothoa distans		Schizotheca fissa	
Tubulipora penicillata			Hippoporina pertusa	
			Alcyonidium variegatum	

Group II. Species common on stone but also commonly present on other supports.

Colony Type

Erect	Spreading	Incrusting: Large	Incrusting: Small	Nodular
Crisidia cornuta	Tubulipora phalangea	Electra pilosa	Diplosolen obelia	Celleporaria pumicosa
Crisia aculeata	Aetea sica	Porella concinna	Diastopora patina	
Crisia denticulata	Pyripora catenularia	Parasmittina trispinosa	Diastopora suborbicularis	
Crisia ramosa	Beania mirabilis	Escharella variolosa	Lichenopora hispida	
Scrupocellaria scruposa	Hippothoa divaricata		Membraniporella nitida	
Scrupocellaria reptans	Alcyonidium mamillatum		Microporella ciliata	
Bugula flabellata	Nolella dilatata		Fenestrulina malusi	
Flustra foliacea			Chorizopora brongniarti	
Cellaria fistulosa			Escharella immersa	
Cellaria sinuosa			Escharella ventricosa	
Alcyonidium gelatinosum			Schizomavella auriculata	
			Schizomavella linearis	

Group III. Species commonly present on stone rare on other supports

Colony Type

Spreading	Spreading	Incrusting: Large	Incrusting: Small
Lepralia foliacea	-	Reptadeonella violacea	Escharoides coccineus
			Schizoporella unicornis
			Alcyonidium mytili

Table 40 . Differences in tolerance to rough surface textures among ectoprocts occurring on shell.

Species confined to or clearly most abundant on smooth surfaces	Species indifferent to surface texture
Diplosolen obelia	*Diastopora patina*
Diastopora suborbicularis	*Electra pilosa*
Lichenopora hispida	*Amphiblestrum flemingi*
Callopora craticula	*Hippoporina pertusa*
Callopora lineata	*Escharella immersa*
Callopora dumerili	*Porella concinna*
Alderina imbellis	*Parasmittina trispinosa*
Microporella ciliata	*Schizomavella auriculata*
Figularia figularis	*Escharoides coccineus*
Cribrilaria radiata	*Alcyonidium variegatum*
Fenestrulina malusi	
Chorhizopora brongniarti	
Escharella ventricosa	
Escharella variolosa	
Schizotheca fissa	
Schizomavella auriculata	
Schizomavella linearis	
Smittina cheilostomata	
Smittina landsborovi	
Smittoidea reticulata	
Alcyonidium mytili	

Table 41. The Ectoprocts occurring on the shores of the Isle of Man.

Rare species	Species rare on shore not uncommon sub-littorally	Species common on shore rare sub-littorally	Species common on shore and not uncommon sub-littorally
Filicrisia geniculata	Crisia aculeata	Membranipora membranacea	Crisidia cornuta
Conopeum reticulum	Crisia denticulata	Callopora aurita	Crisia eburnea
	Aetea anguinea	Caulorampus spiniferum	Tubulipora phalangea
	Aetea sica	Haplopoma graniferum	Diplosolen obelia
	Bicellariella ciliata	Cryptosula pallasiana	Diastopora patina
	Beania mirabilis	Umbonula littoralis	Diastopora suborbicularis
	Callopora dumerili	Escharina spiniforum	Lichenopora hispida
	Amphiblestrum flemingi	Alcyonidium hirsutum	Scruparia ambigua
	Chorizopora brongniarti	Alcyonidium polyoum	Scruparia chelata
	Escharella ventricosa	Flustrellidra hispida	Scrupocellaria reptans
	Schizomavella auriculata	Bowerbankia imbricata	Electra pilosa
	Parasmittina trispinosa	Valkeria uva	Callopora lineata
	Alcyonidium gelatinosum		Cribrilina punctata
	Alcyonidium mammillatum		Membraniporella nitida
			Hippothoa hyalina
			Microporella ciliata
			Escharella immersa
			Schizoporella unicornis
			Schizomavella linearis
			Escharoides coccineus
			Celleporaria pumicosa
			Celleporina costazii
			Alcyonidium mytili
			Bowerbankia gracilis

Table 42. Reproductive habits and distribution of Manx ectoprocts.

	Species with a large proportion of the zooids in each colony fertile			Species in which few zooids in each colony are fertile		
	Distribution			Distribution		
	Offshore		Shore	Offshore		Shore
	Limited	Wide		Limited	Wide	
Long reproductive season	4	14	-	5	2	-
Short reproductive season	3	10	12	-	2	-

Excluding species with cyphonautes larvae and cyclostome species

Table 43. Reproductive habits and distribution of shore ectoprocts
Only species common on the shore and rare elsewhere are included.

Distribution	Species brooding embryos in ooecia	Species brooding embryos internally
No. of species in exposed positions	-	7
No. of species in sheltered positions	5	1

Exposed and sheltered refer to the exposure to dessiccating influences.

Table 44 . Seasons of peak reproduction of 55 ectoproct species in Manx waters.

Season	No. of species with peak in each season of			
	25 species with embryos all year	10 species with embryos for 9-11 months	11 species with embryos for 6-8 months	9 species with embryos for less than 6 months
Sp.	1	-	1	2
Su.	5	2	3	2
Au.	-	-	1	1
Wn.	1	1	1	1
Sp-Su.	3	1	2	-
Su-Au.	2	-	1	1
Au-Wn.	2	3	2	1
Wn-Sp.	-	1	-	-
Sp. and Au.	-	-	-	1
Au-Wn+Sp.	1	-	-	-
Su-Au-Wn	2	1	-	-
No peak	8	1	-	-

Sp = Spring= May, June, July
Su= Summer= August, September, October
Au = Autumn= November, December, January.
Wn= Winter= February, March, April.

These seasons are based on sea-temperature (See Fig. II 77)

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Table 45 . Number of ectoproct species reproducing in each m
at Provence (data from Gautier 1962) , in the English Channel
(Plymouth and Roscoff combined. Data from P.M.F. (1957) and
Echalier and Prenant (1951.)

Locality		Month and no. of species reproducing									
		J	F	M	A	M	J	J	A	S	O
PROVENCE (Cheilostomata only)		17	19	18	25	33	33	27	31	37	34
CHANNEL		-	-	-	1	3	5	6	18	23	25
ISLE	Cheilostomata	43	38	42	38	40	40	38	45	51	47
OF	Ctenostomata	5	6	6	4	3	4	2	6	5	6
MAN	Cyclostomata	3	3	5	6	3	7	4	3	5	5
	Total	51	47	53	48	46	51	54	54	61	58

Table 46 . Number of species for which full data on reproduction was not obtained reproducing in each month grouped according to geographical distribution.

13 species occurring in Arctic and either Mediterranean or Tropics or both

	Season and Month											
	Winter			Spring			Summer			Autumn		
	F	M	A	M	J	J	A	S	O	N	D	J
No. of species recorded as reproducing	4	5	6	2	8	3	4	3	5	5	5	3

14 species occurring in Mediterranean and Tropics but not Arctic

	Season and Month											
	Winter			Spring			Summer			Autumn		
	F	M	A	M	J	J	A	S	O	N	D	J
No. of species recorded as reproducing	2	3	1	3	4	1	5	7	6	6	3	3

2 species occurring in Arctic and not Mediterranean or Tropics.

	Season and Month											
	Winter			Spring			Summer			Autumn		
	F	M	A	M	J	J	A	S	O	N	D	J
No. of species recorded as reproducing	1							1			1	

Seasons are based on sea temperatures (See fig II 77)

Table 47. Summary of the data on reproductive season of those species from which full data on reproductive season has been obtained, grouped according to geographical distribution of the species included.

24 Species from Arctic and Mediterranean of Tropics or both

	Reproductive Season							
	All year	WnSpSu	SpSu	Su	Au	AuWn	AuWnSp.	WnSp
No. of species	15	1	1	3	1	1	1	1

19 Species from Mediterranean or Tropics but not Arctic.

	Reproductive Season				
	All year	SpSu	Su	SpSuAu	AuWnSp
No. of species	13	1	2	2	1

5 Species from Arctic but not Mediterranean or Tropics.

	Reproductive Season			
	Sp.	Au.	AuWn.	Wn.
No. of species	1	1	1	2

Season of peak reproduction in 15 species from Arctic and Mediterranean or Tropics or both which reproduce all year (see above)

	Season of peak reproduction					
	Sp	Su	SuAu	SuAuWn	AuWn	no obvious peak
No. of species	1	3	1	2	3	5

Season of peak reproduction in 13 species from Mediterranean or Tropics but not Arctic which reproduce throughout the year.

	Season of peak reproduction				
	SpSu	Su	AuWn	AuWnSp	no obvious peak
No. of species	3	4	1	1	4

Sp = May to July
Su = August to October
Au = November to January
Wn = February to April.

Seasons based on sea-temperatures (see fig. II 77.)

Figures 1-77

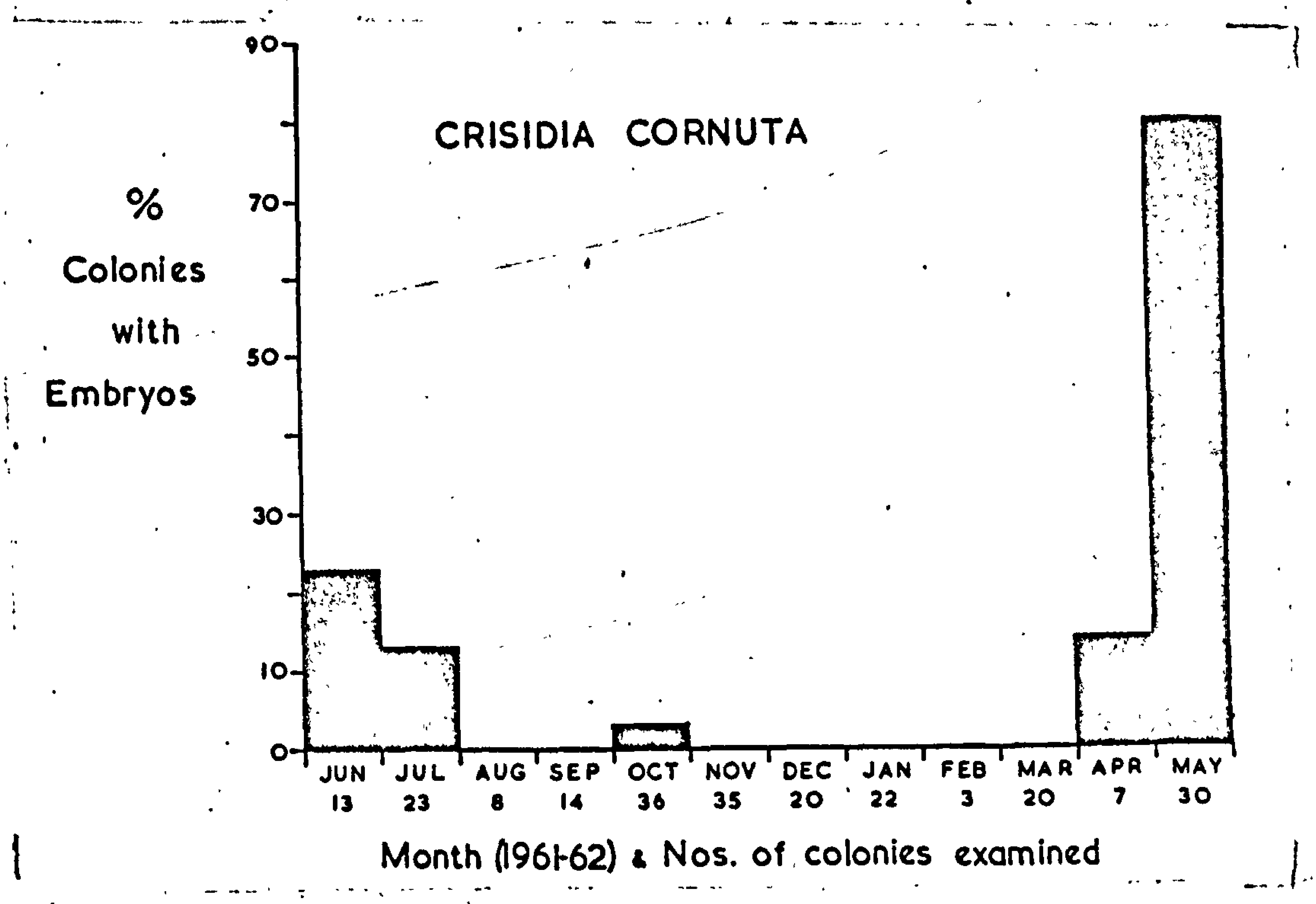


Fig II. 1. Crisidia cornuta. Percentage colonies with embryos each month from June 1961 to May 1962.

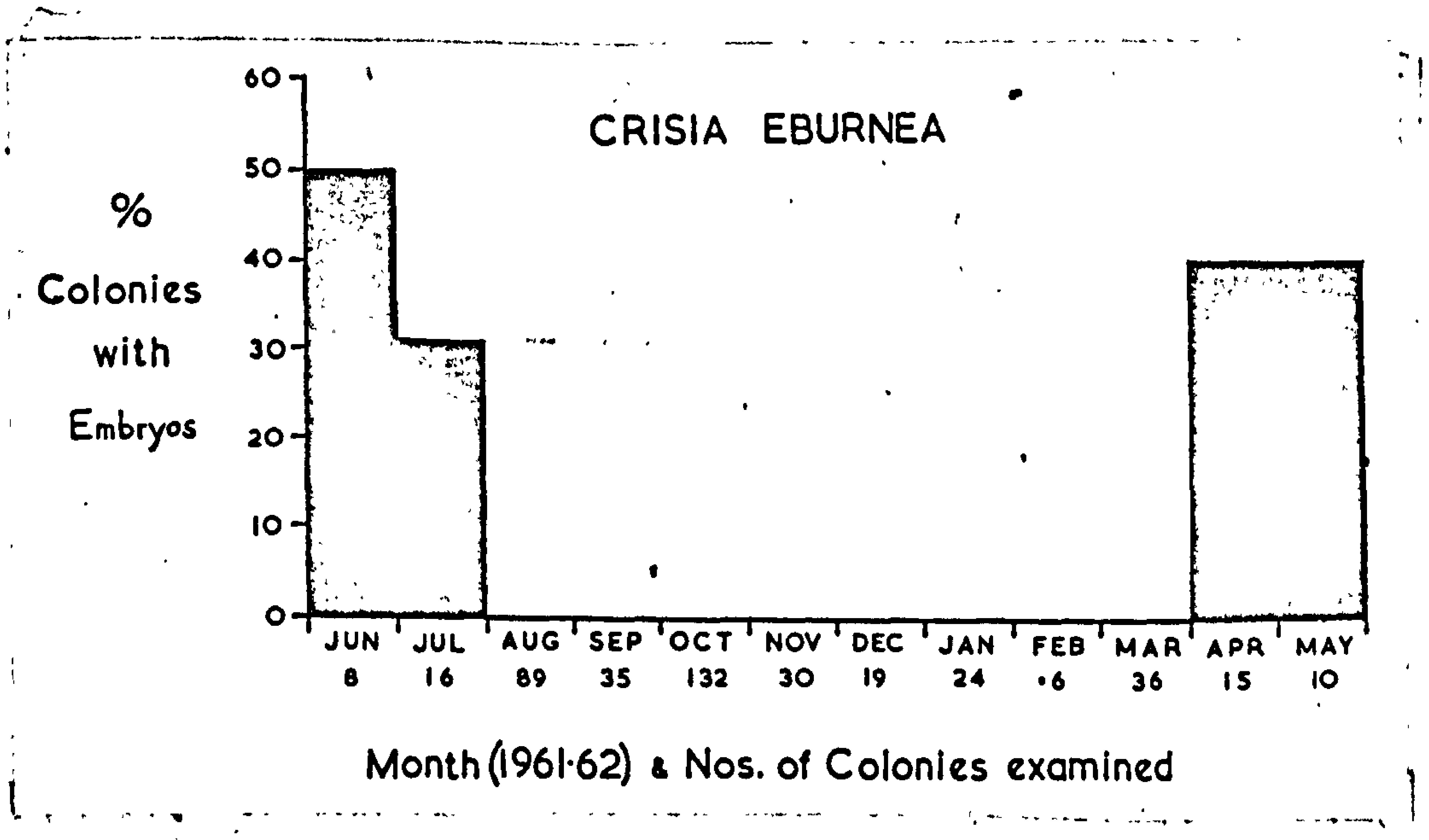


Fig II. 2. Crisia eburnea. Percentage colonies with embryos each month from June 1961 to May 1962.

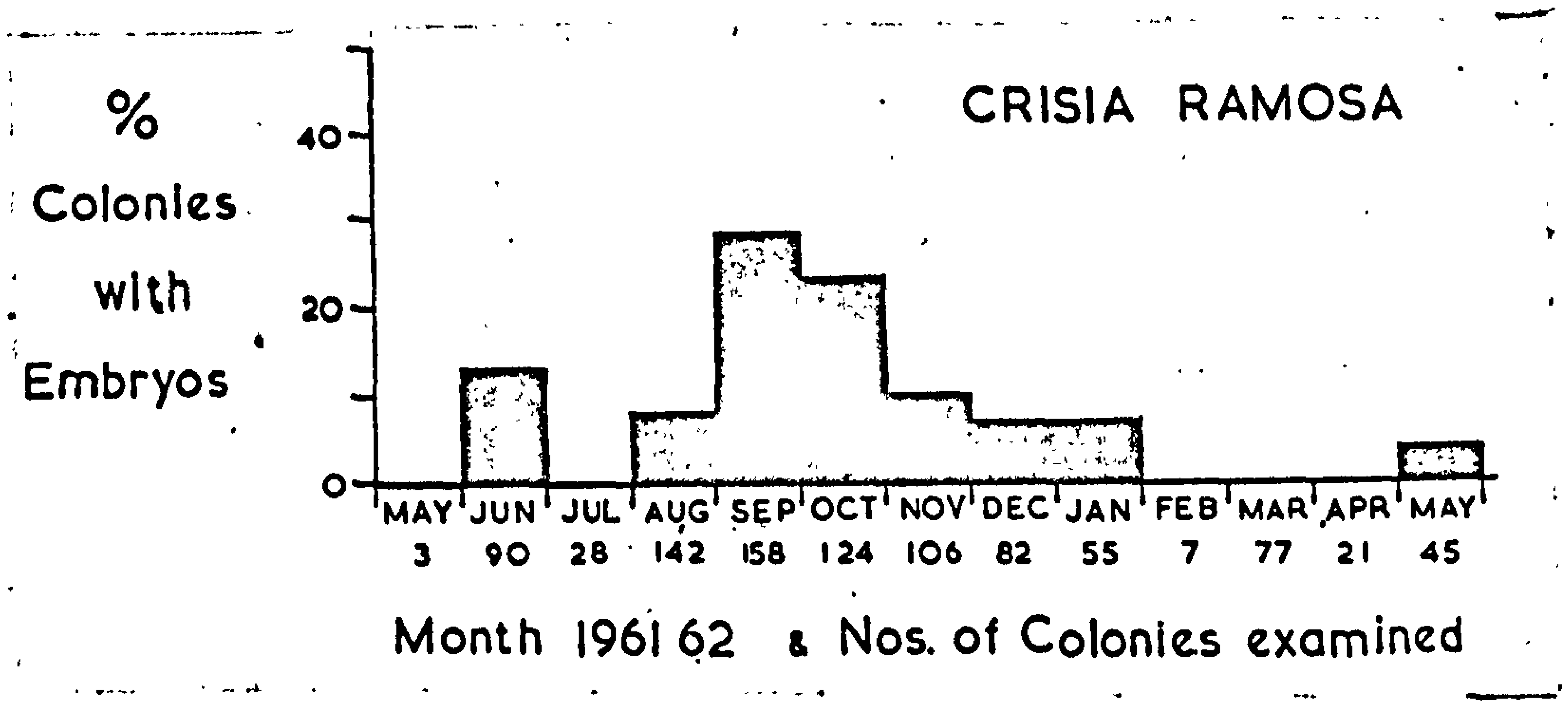


Fig II.3. Crisia ramosa. Percentage colonies with embryos each month from June 1961 to May 1962.

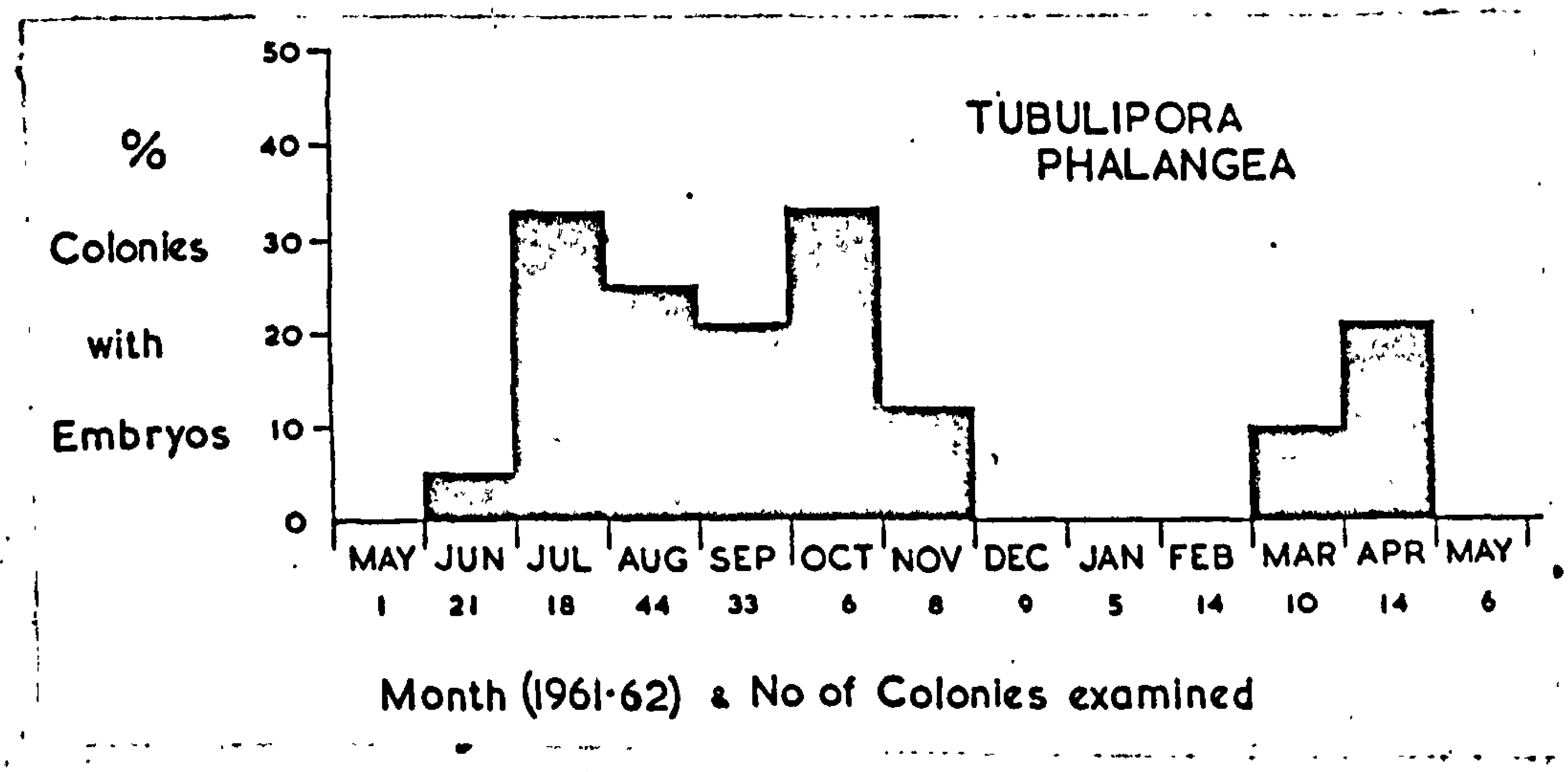


Fig II. 4. Tubulipora phalangea. Percentage colonies with embryos each month from May ;961 to May 1962

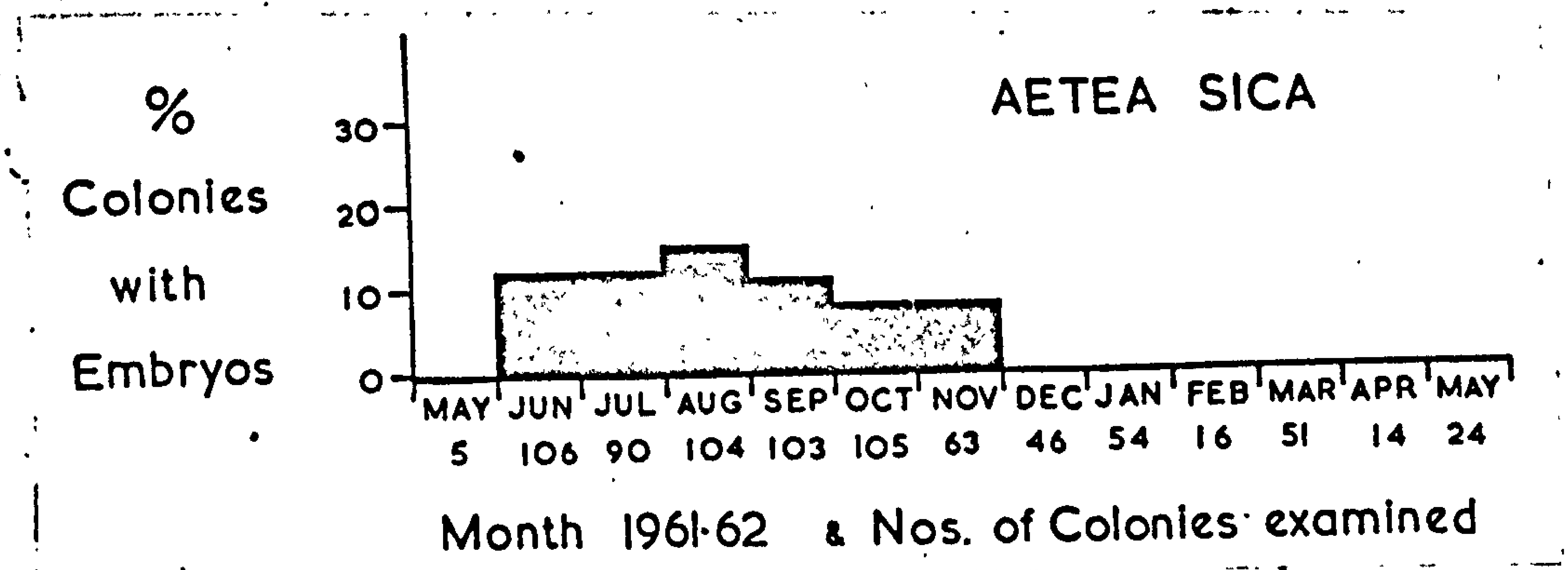


Fig II. 5. Aetea sica. Percentage colonies with embryos in each month's samples from May 1961 to May 1962.

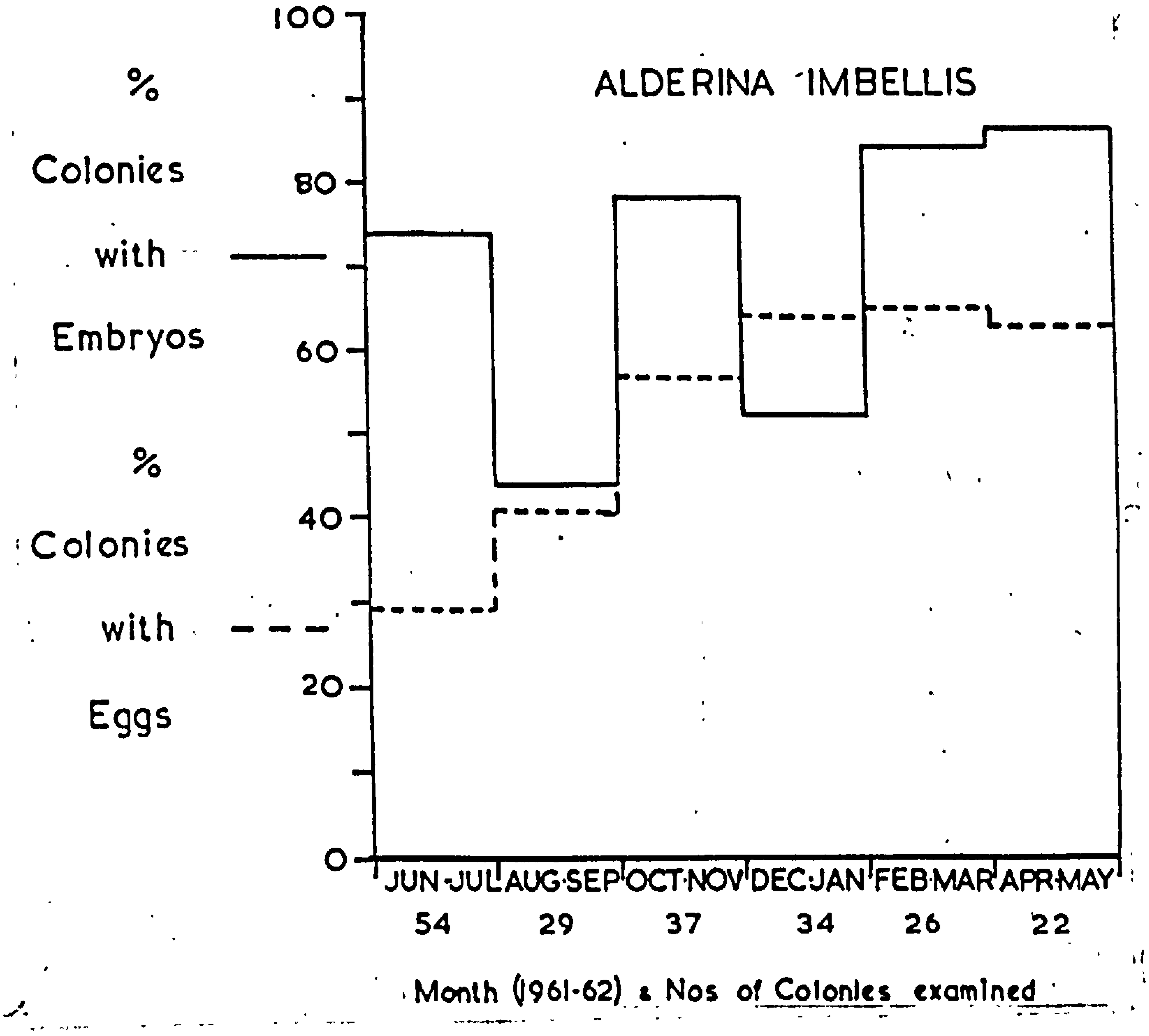


Fig II. 6. Alderina imbellis. Percentage colonies with embryos and percentage colonies with eggs in each two month's samples from June 1961 to May 1962.

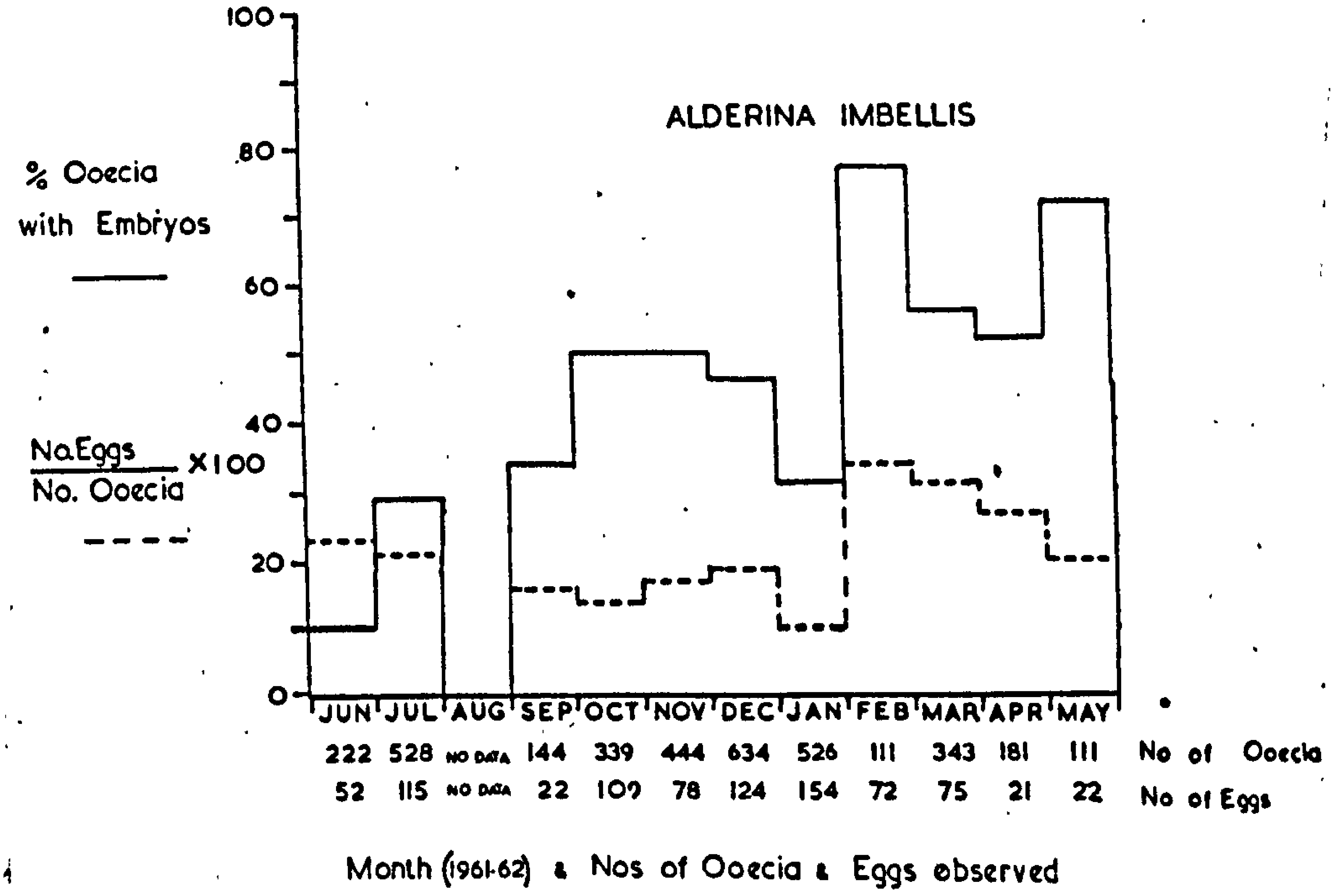


Fig II.7. Alderina imbellis. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia) x 100 in each months samples from June 1961 to May 1962

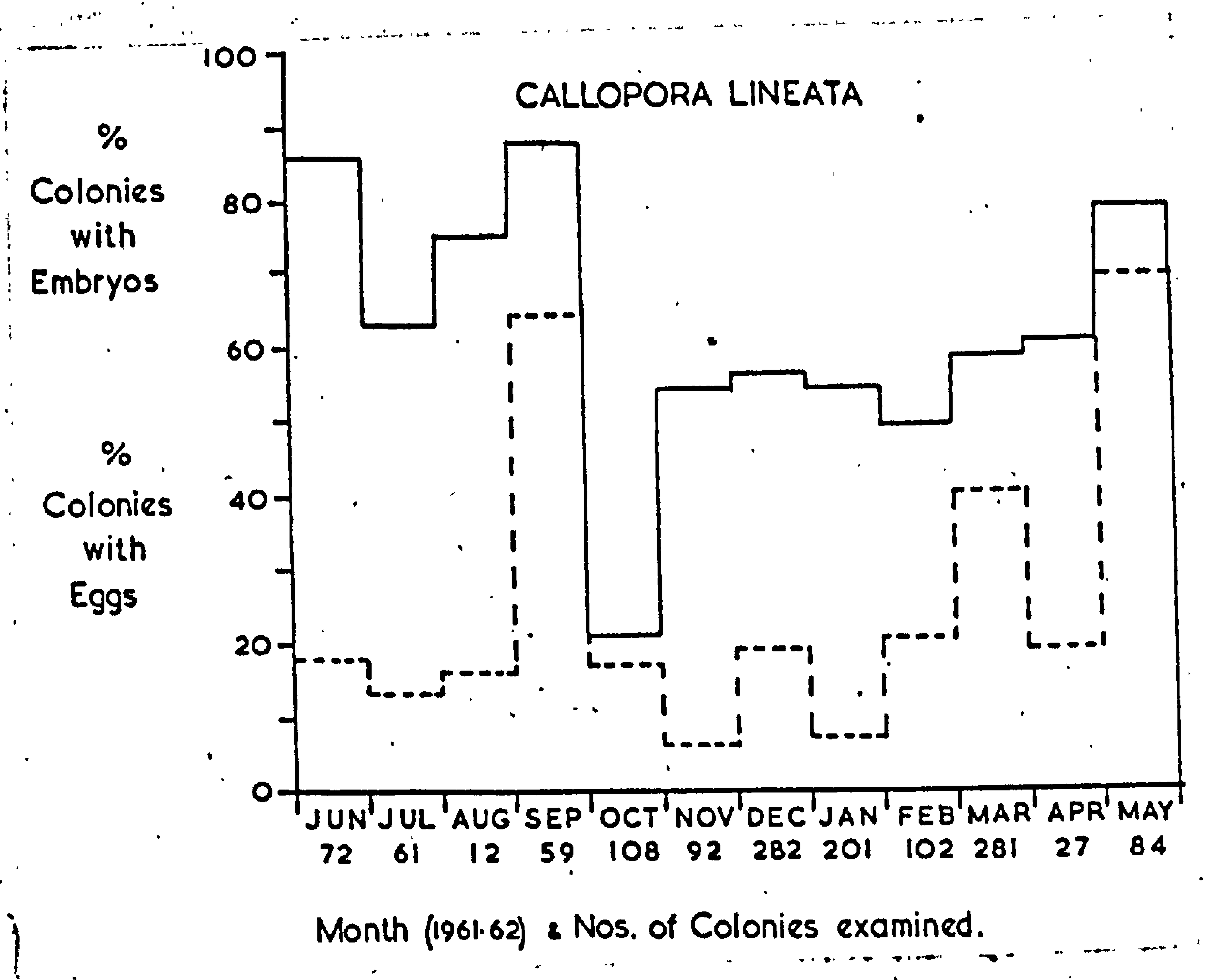


Fig II. 8. Callopora lineata. Percentage colonies with embryos and percentage with eggs in each month's samples between June 1961 and May 1962.

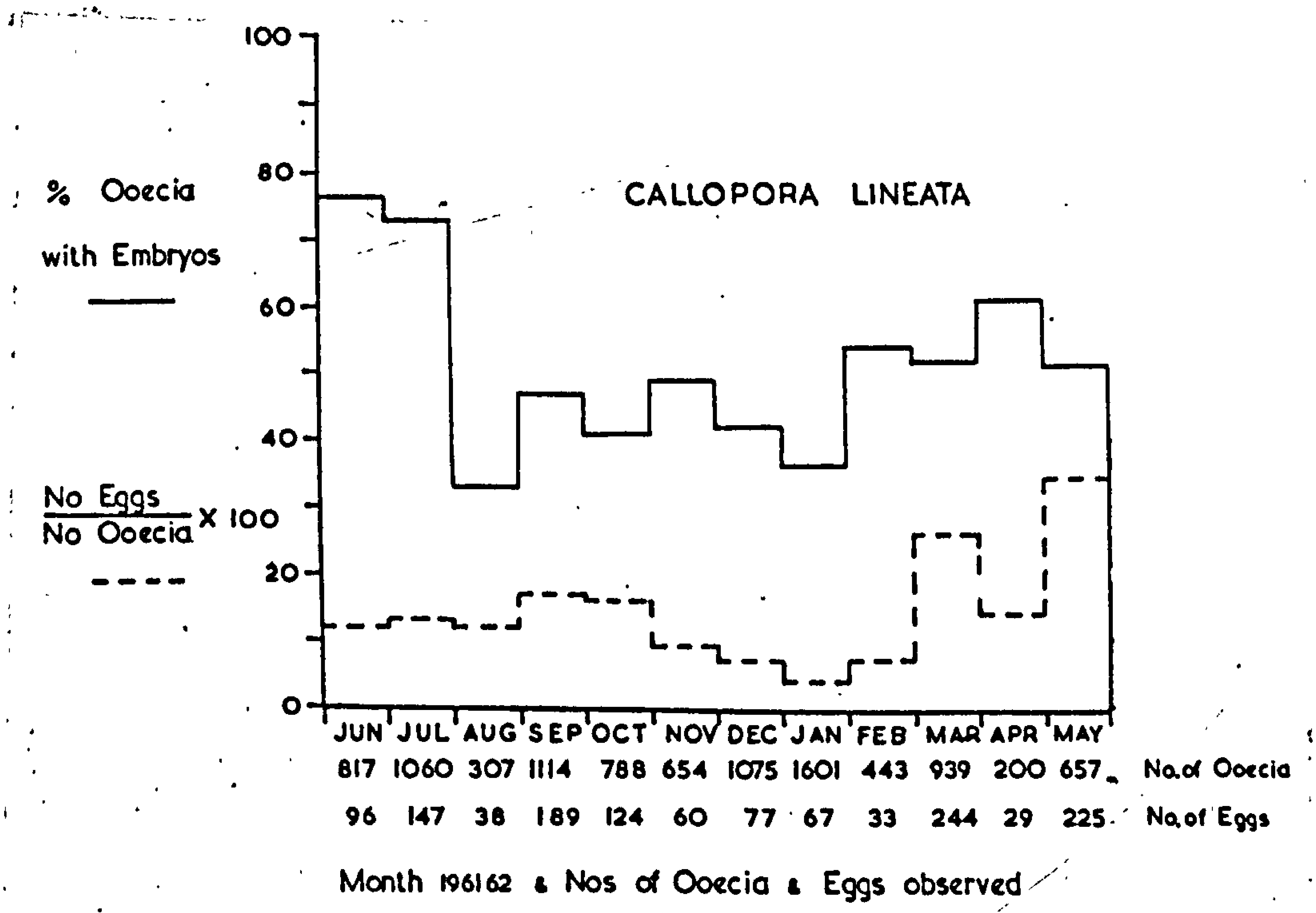


Fig II. 9. Callopora lineata. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia) x 100 in each month's samples from June 1961 to May 1962.

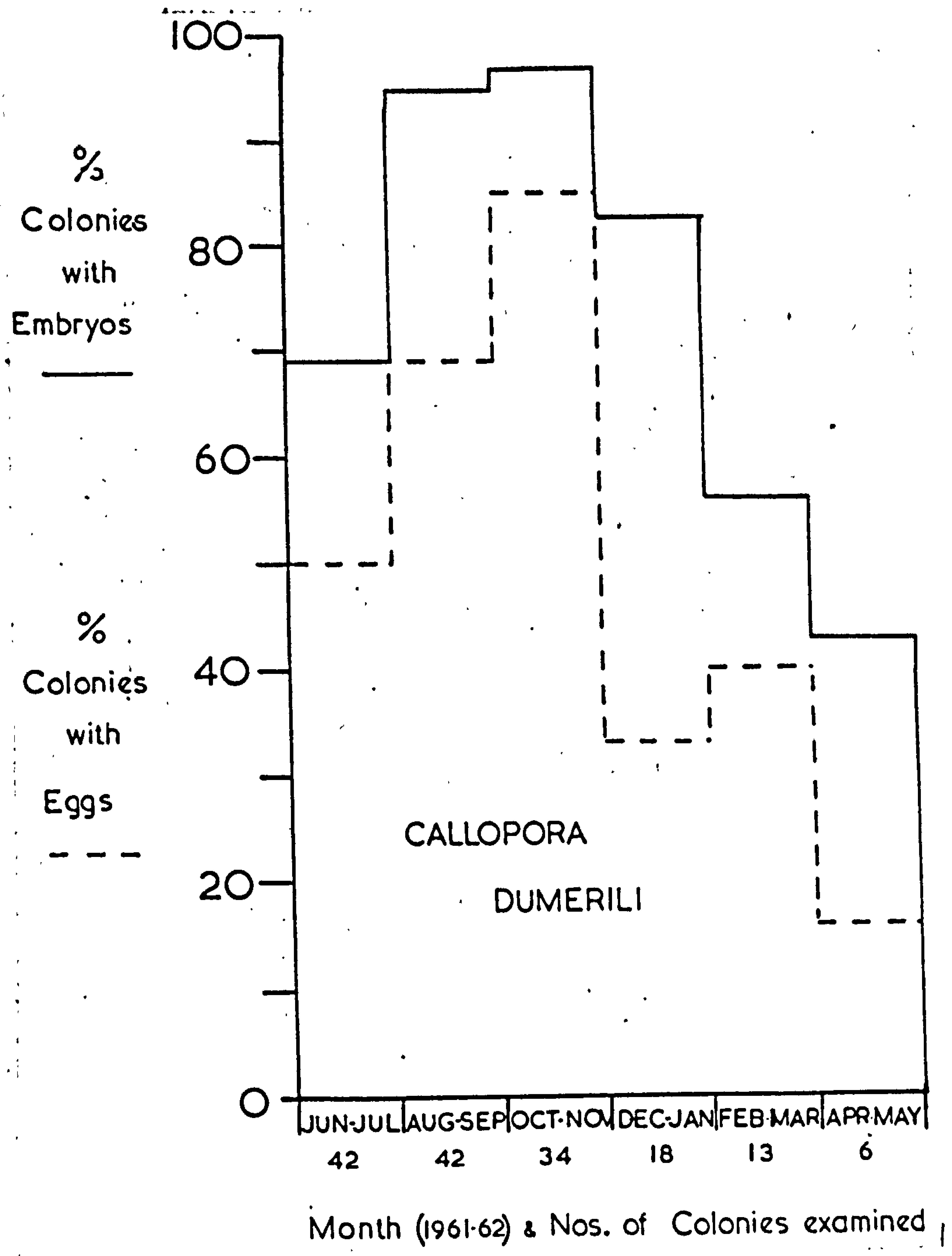


Fig II. 10. Callopora dumerili. Percentage colonies with embryos and percentage with eggs in each two month's samples from June 1961 to May 1962.

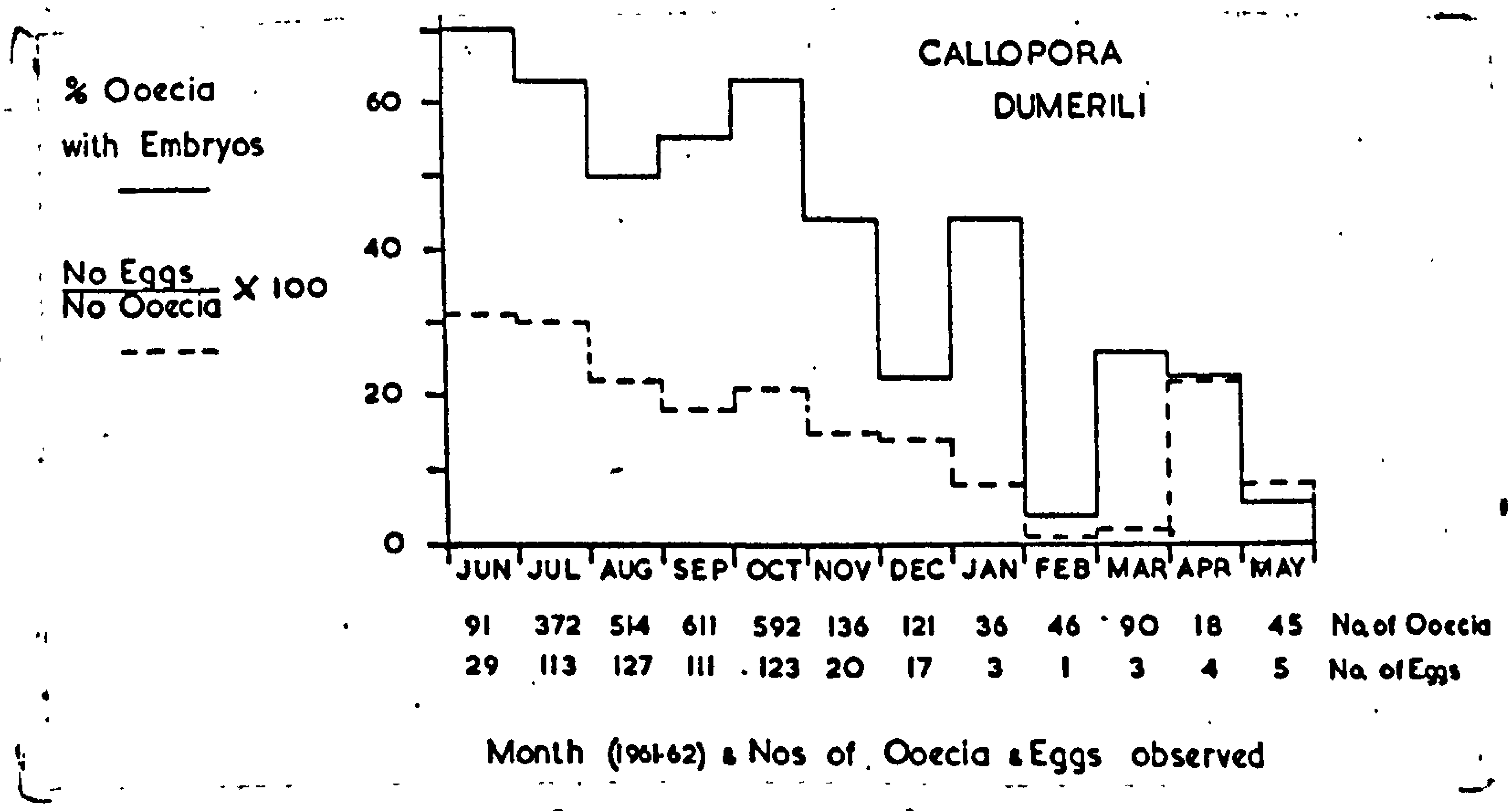


Fig.II.11. Callopora dumerili. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia) ×100 in each month's samplesfrom June 1961 to May 1962

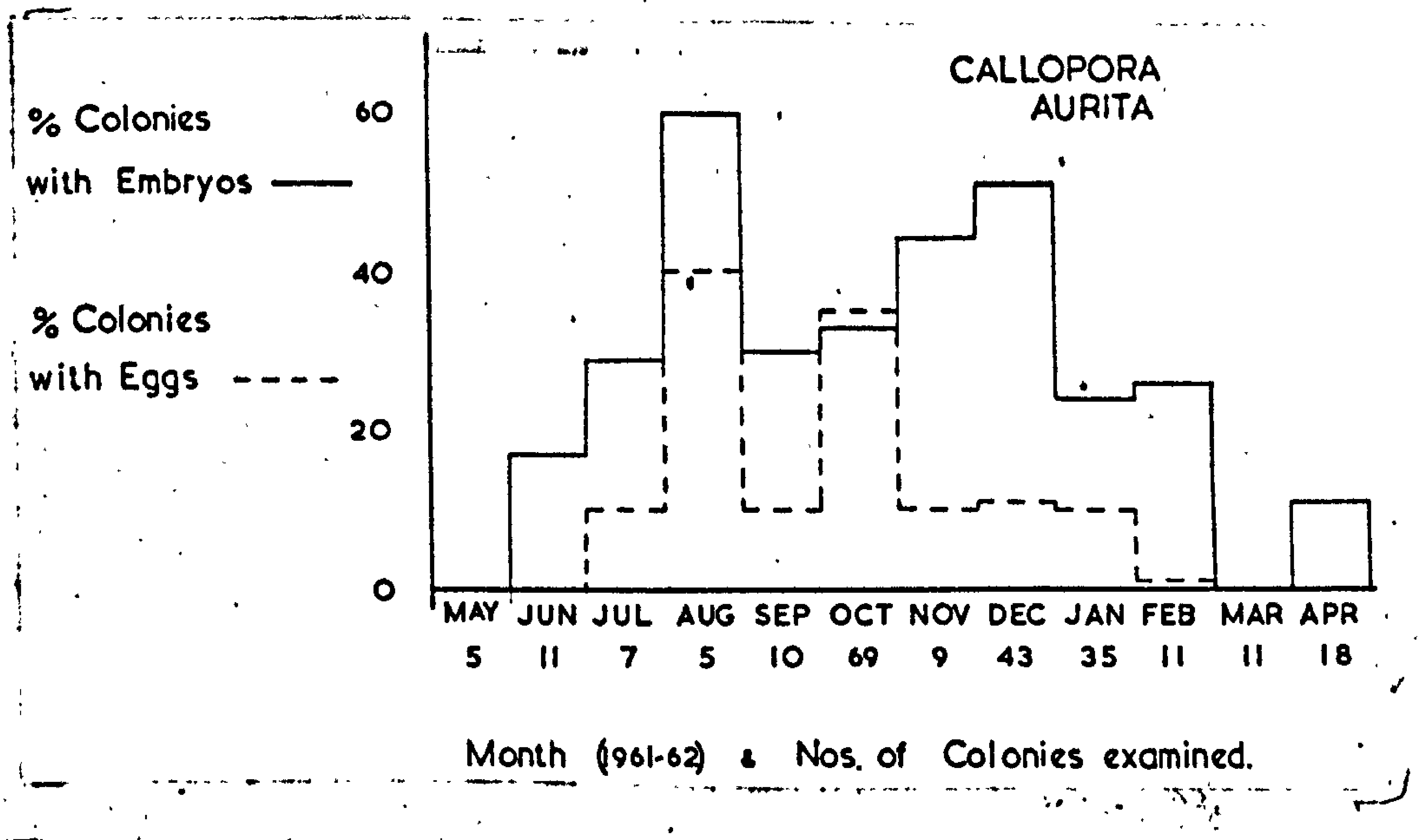


Fig II.12. Callopora aurita. Percentage colonies with embryos and percentage with eggs in each month's samples from May 1961 to April 1962.

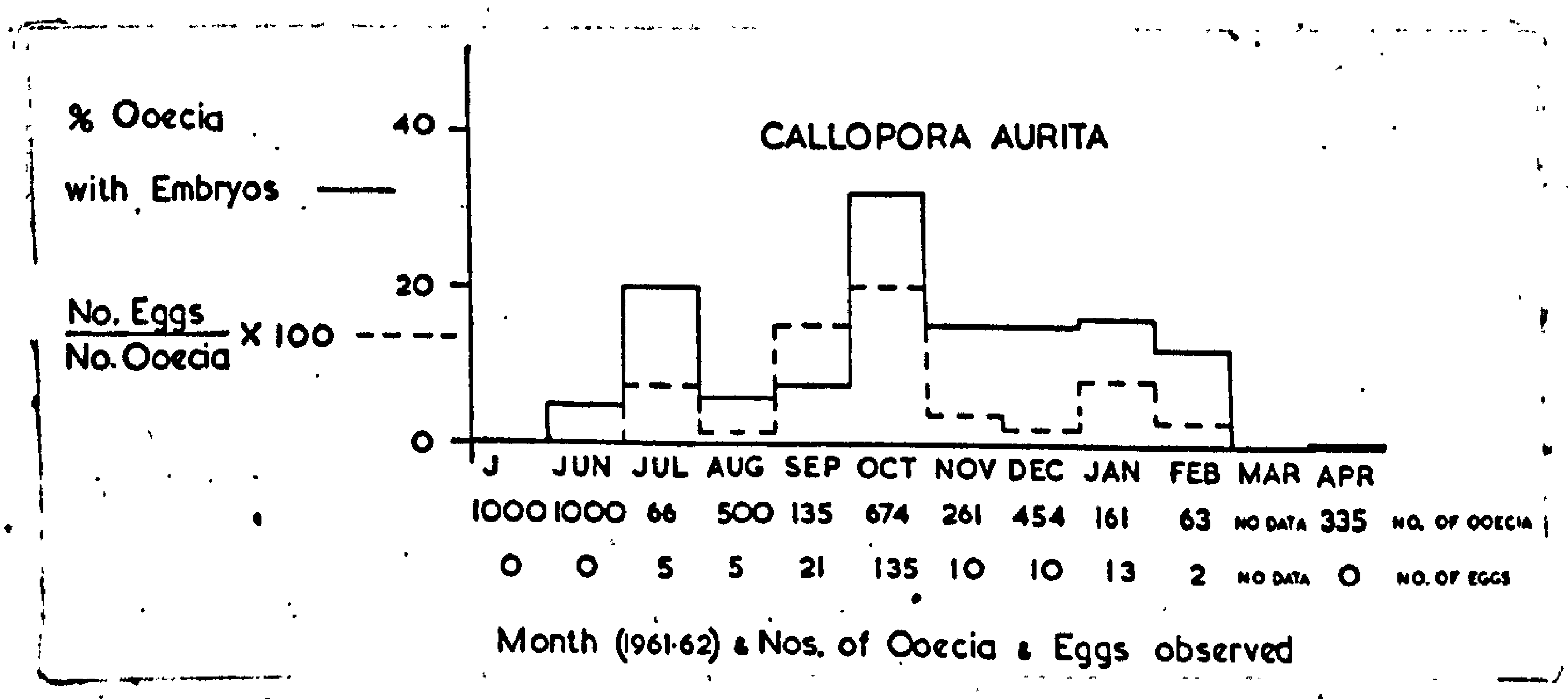


Fig II.13. Callopora aurita. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia)×100 for each month's samples from May 1961 to April 1962.

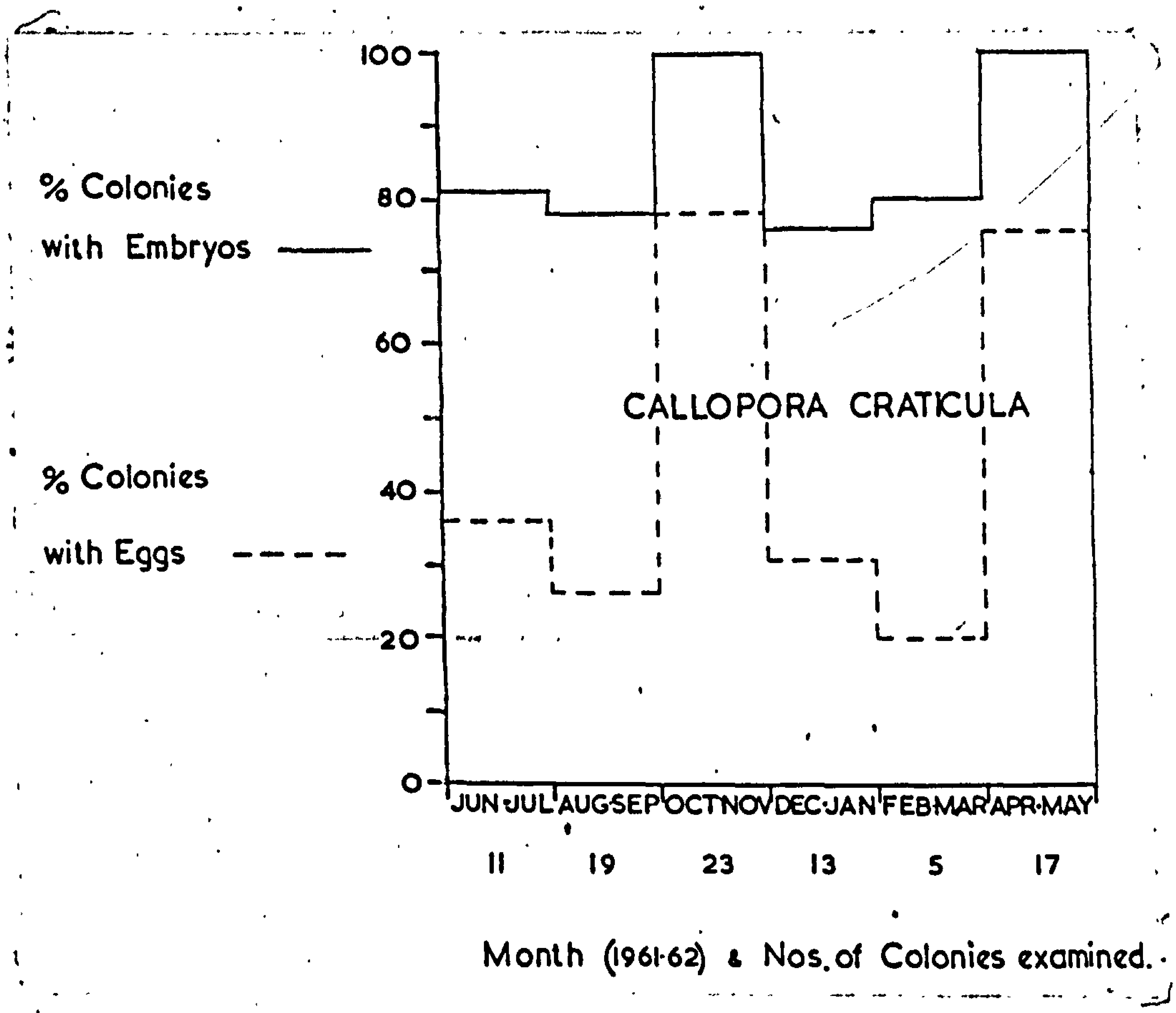


Fig II. 14. Callopora craticula . Percentage of colonies with embryos and percentage with eggs in each two month's samples from June 1961 to May 1962.

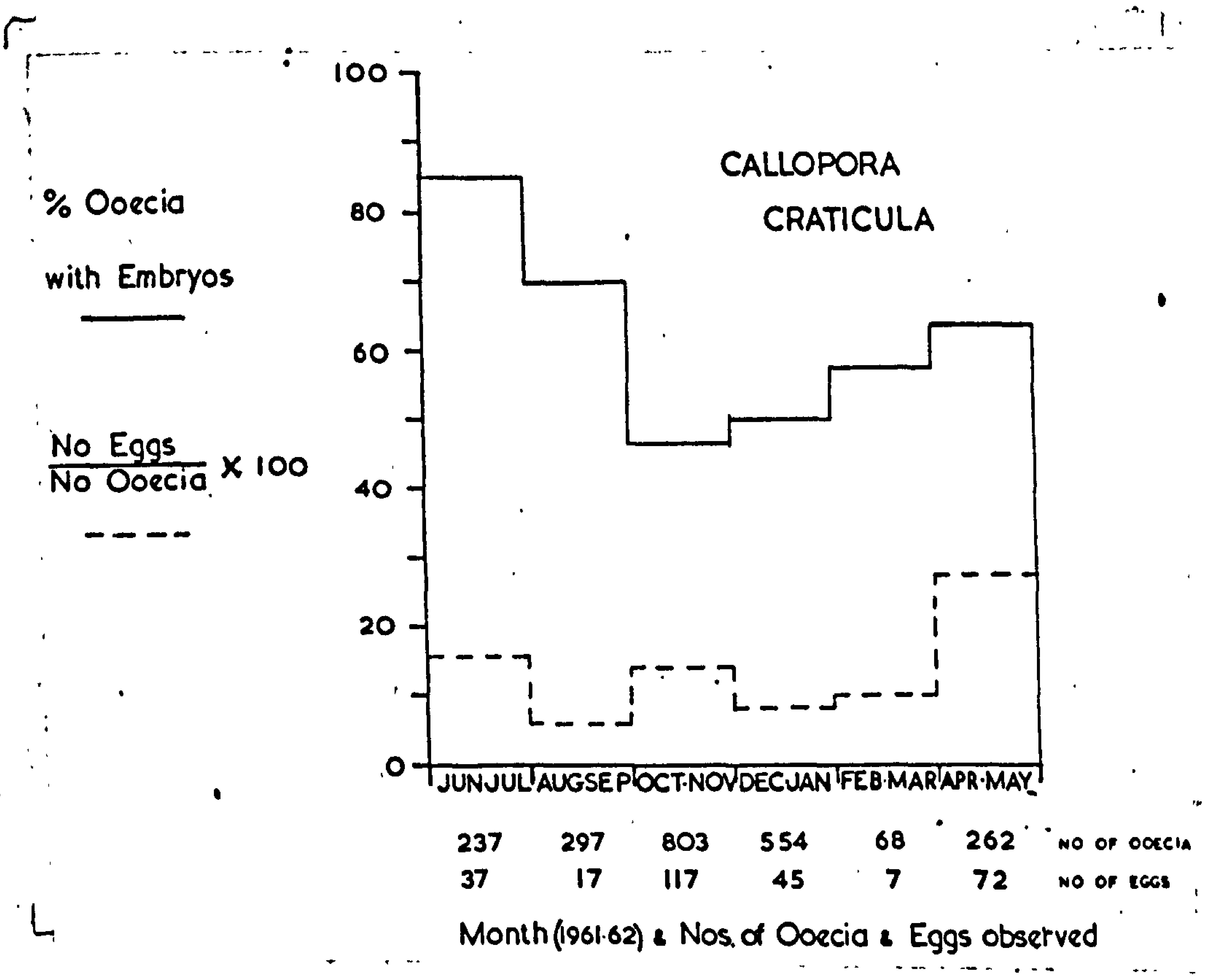


Fig II. 15. Callopora craticula. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia)x 100 for each two months samples from June 1961 to May 1962.

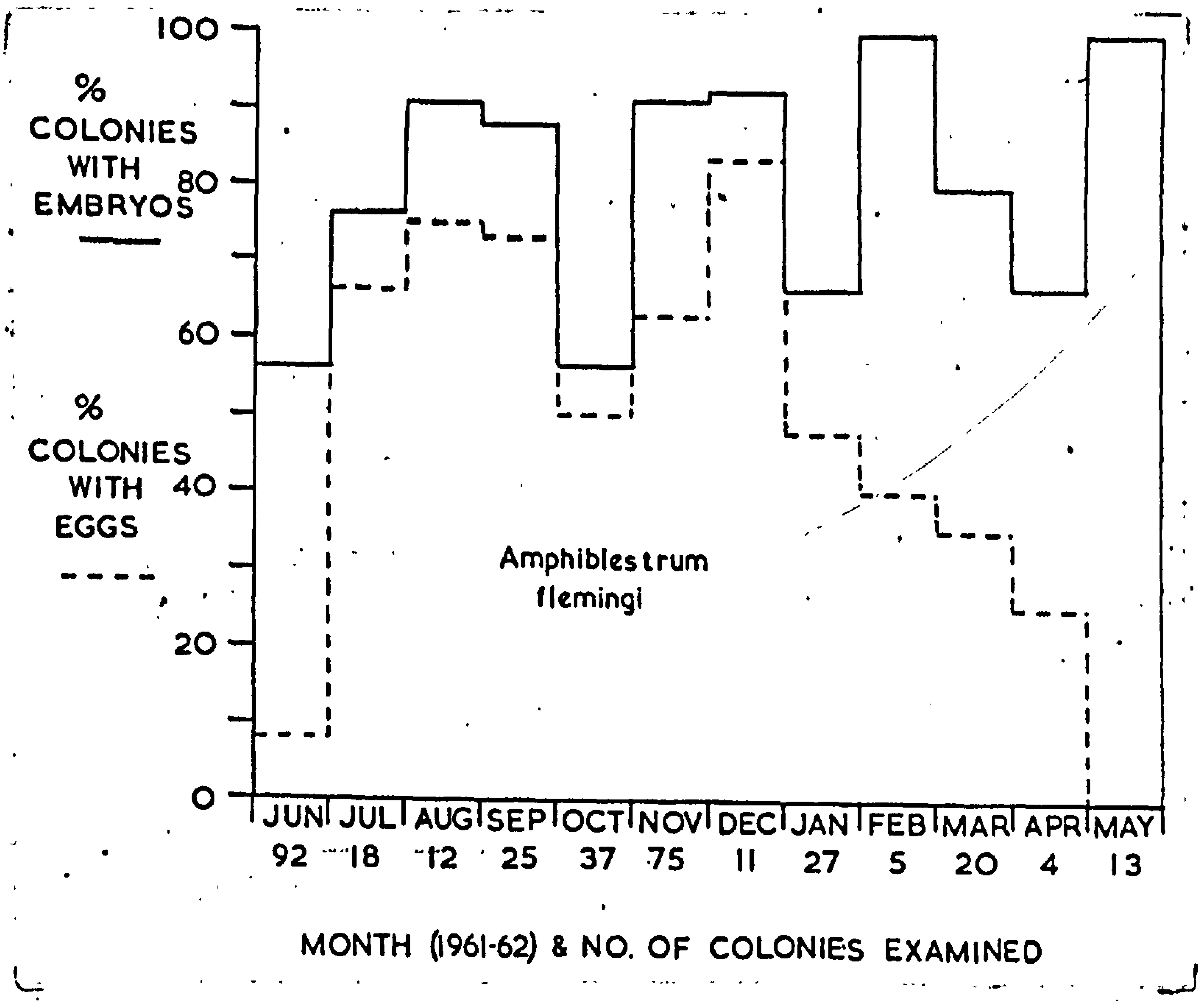


Fig II. 16. Amphiblestrum flemingi. Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May 1962.

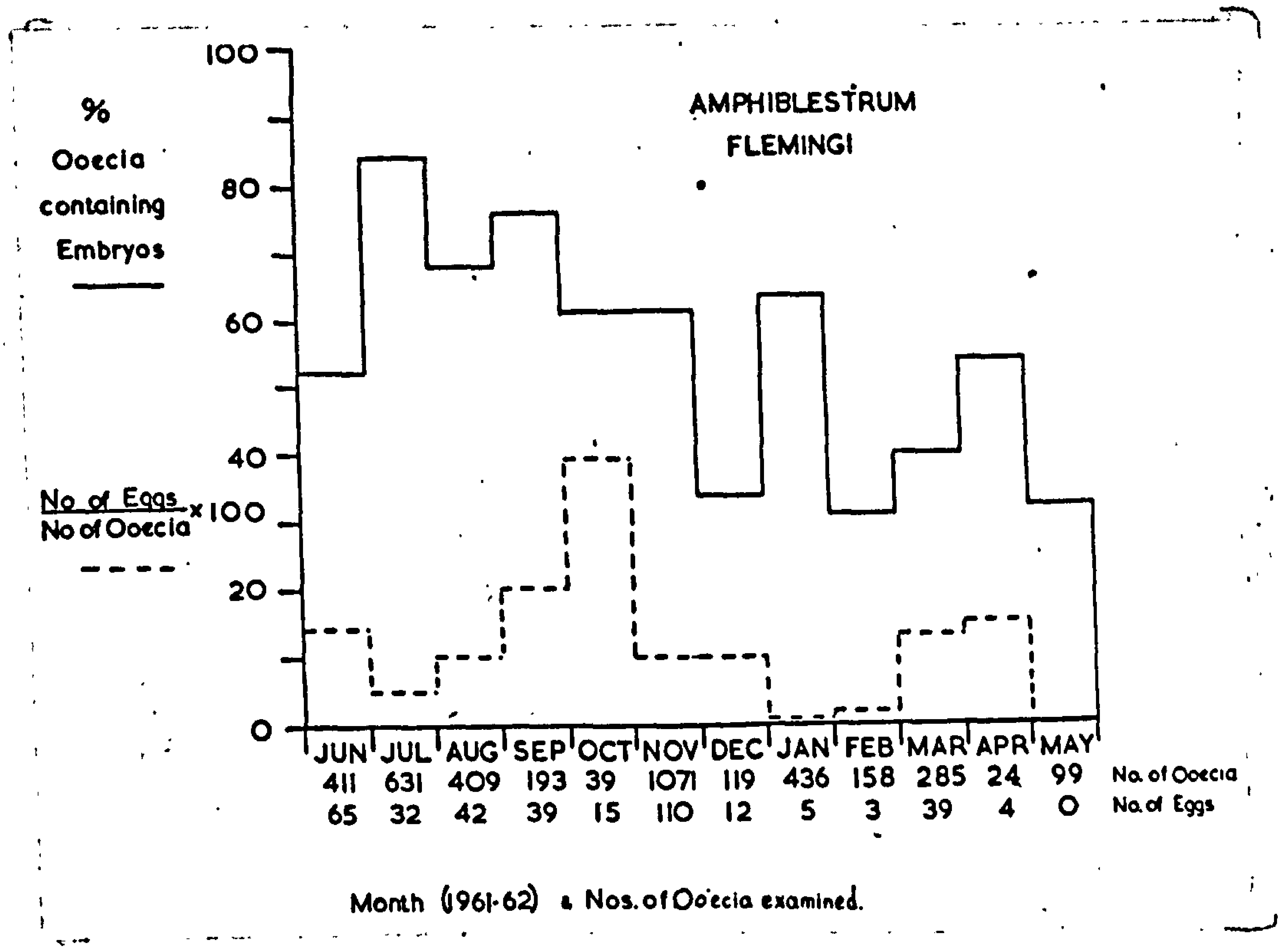


Fig II.17. Amphiblestrum flemingi. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia) x 100 for each month's samples from June 1962 to May 1962

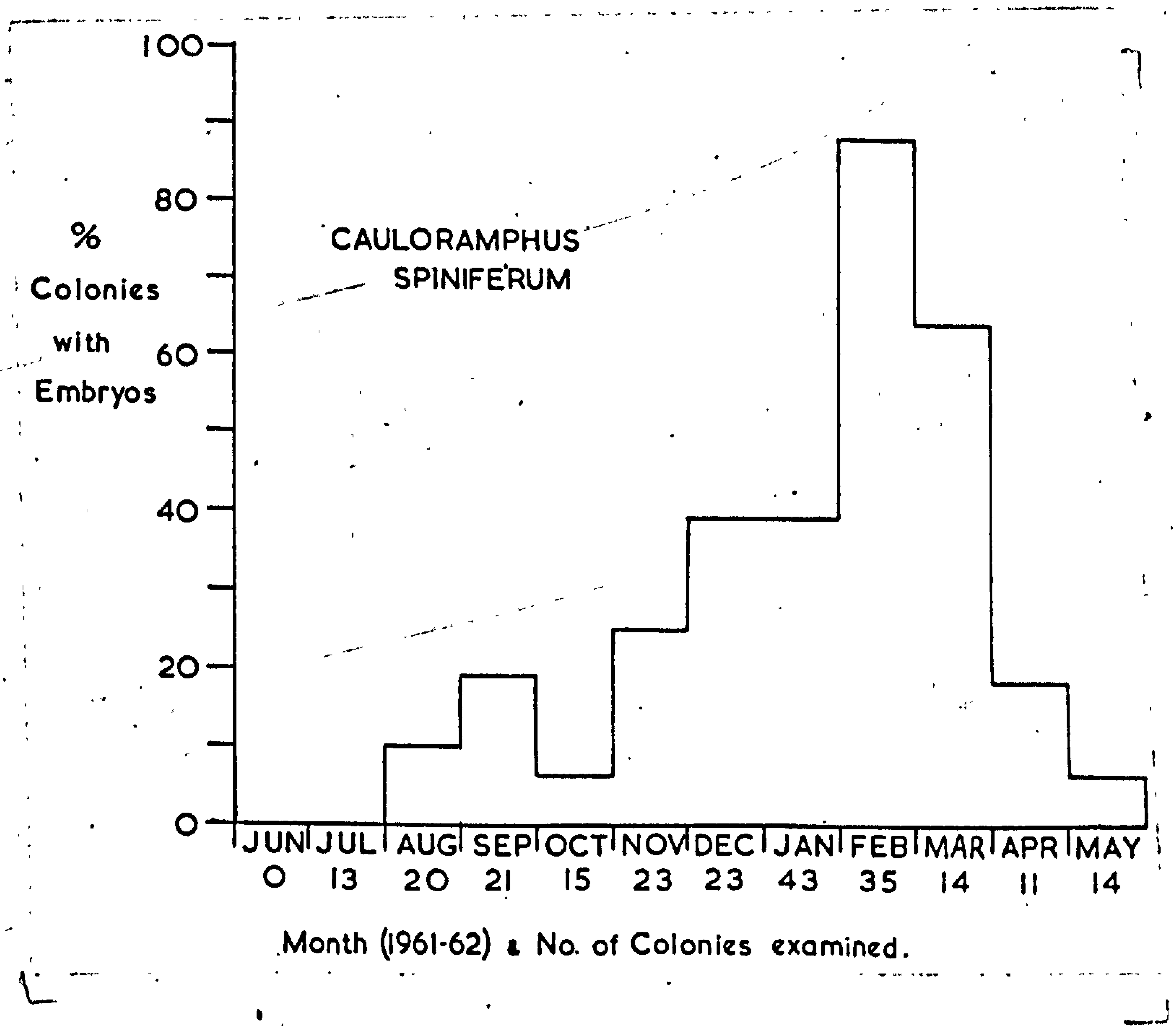


Fig. II.18. Cauloramphus spiniferum. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

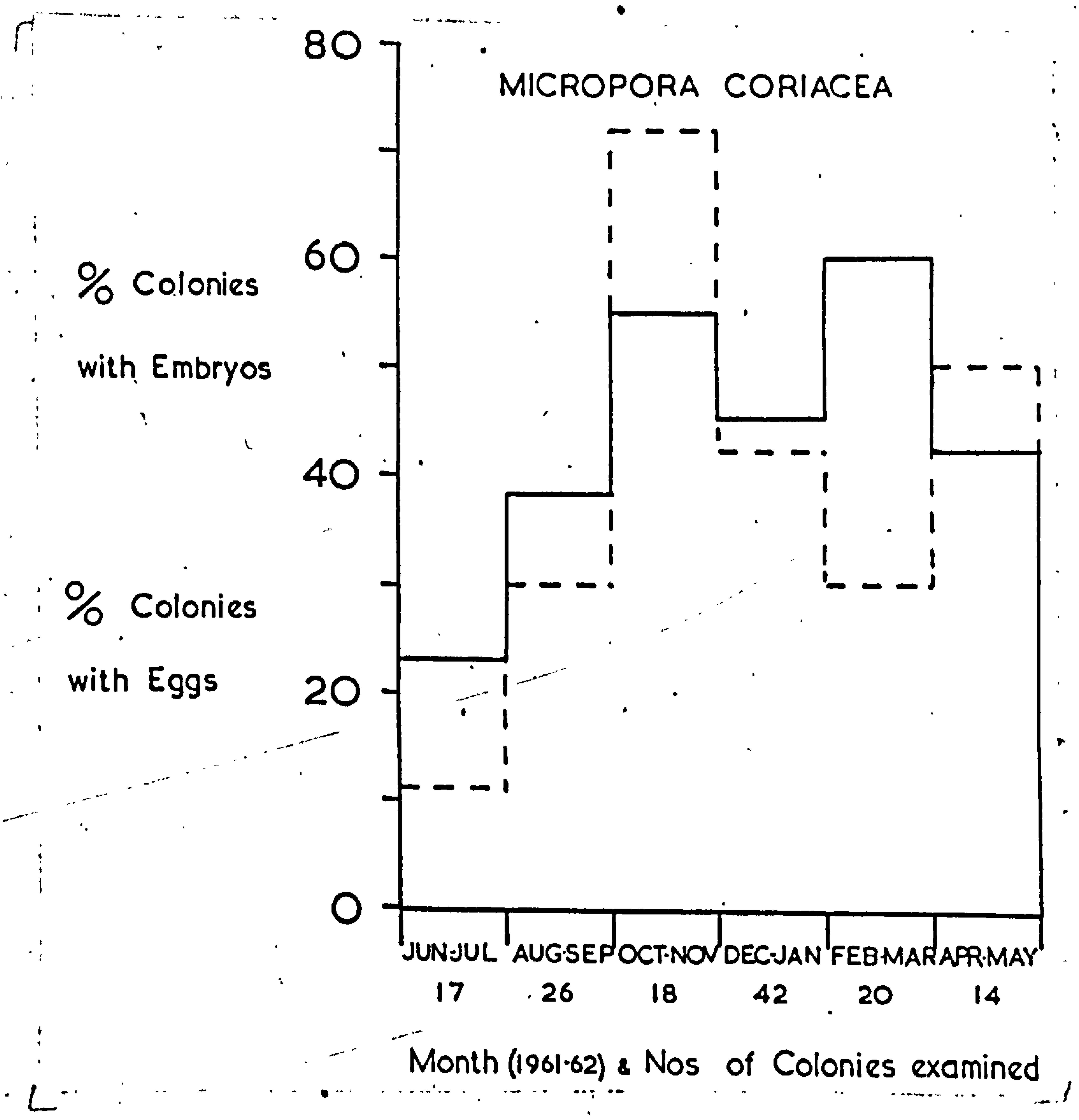


Fig. II.19. Micropora coriacea. Percentage colonies with embryos and percentage with eggs in each two month's samples from June 1961 to May1962.

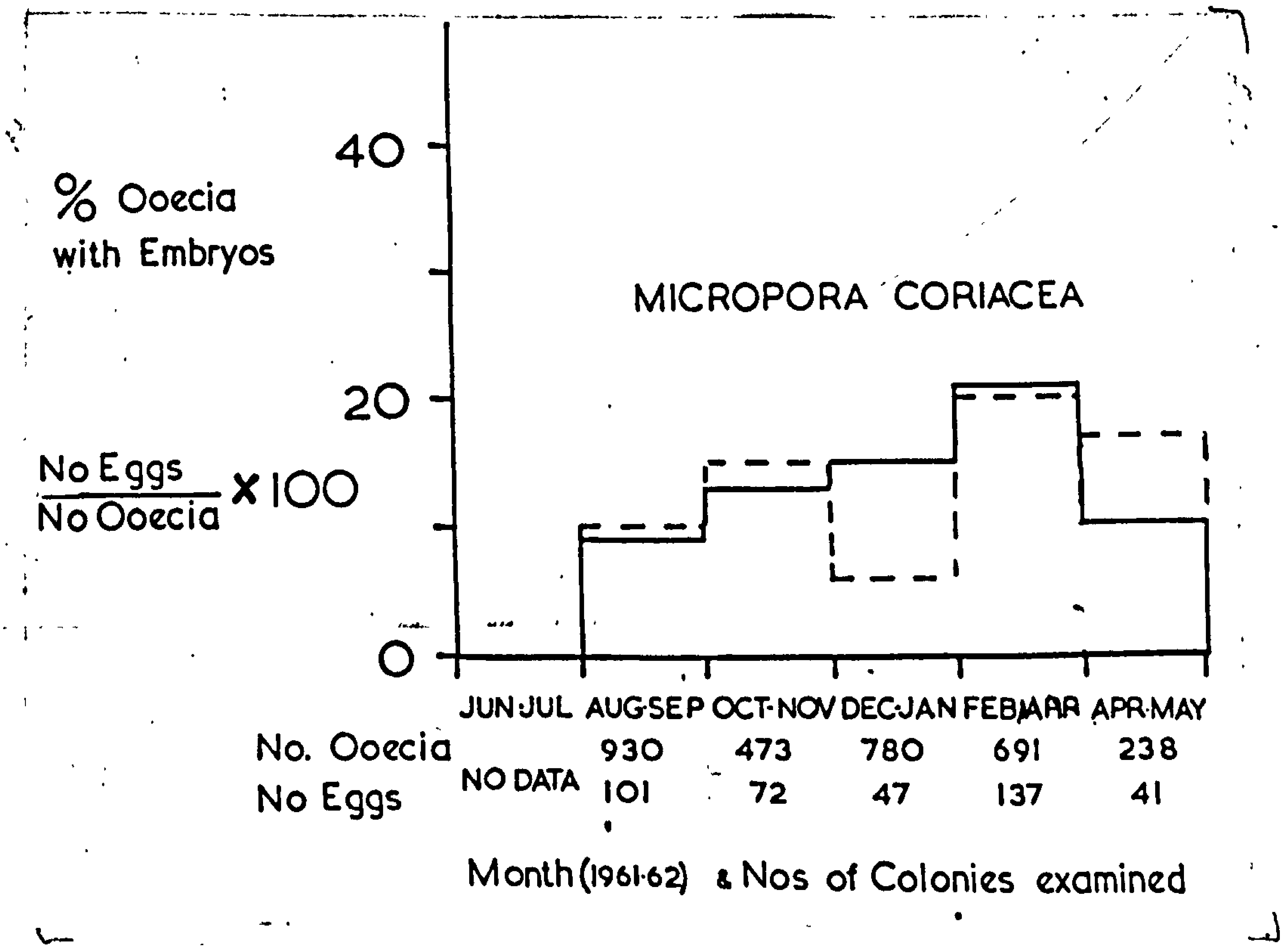


Fig. II. 2o. Micropora coriacea. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia)x 100 in each two month's samples from June 1961 to May 1962

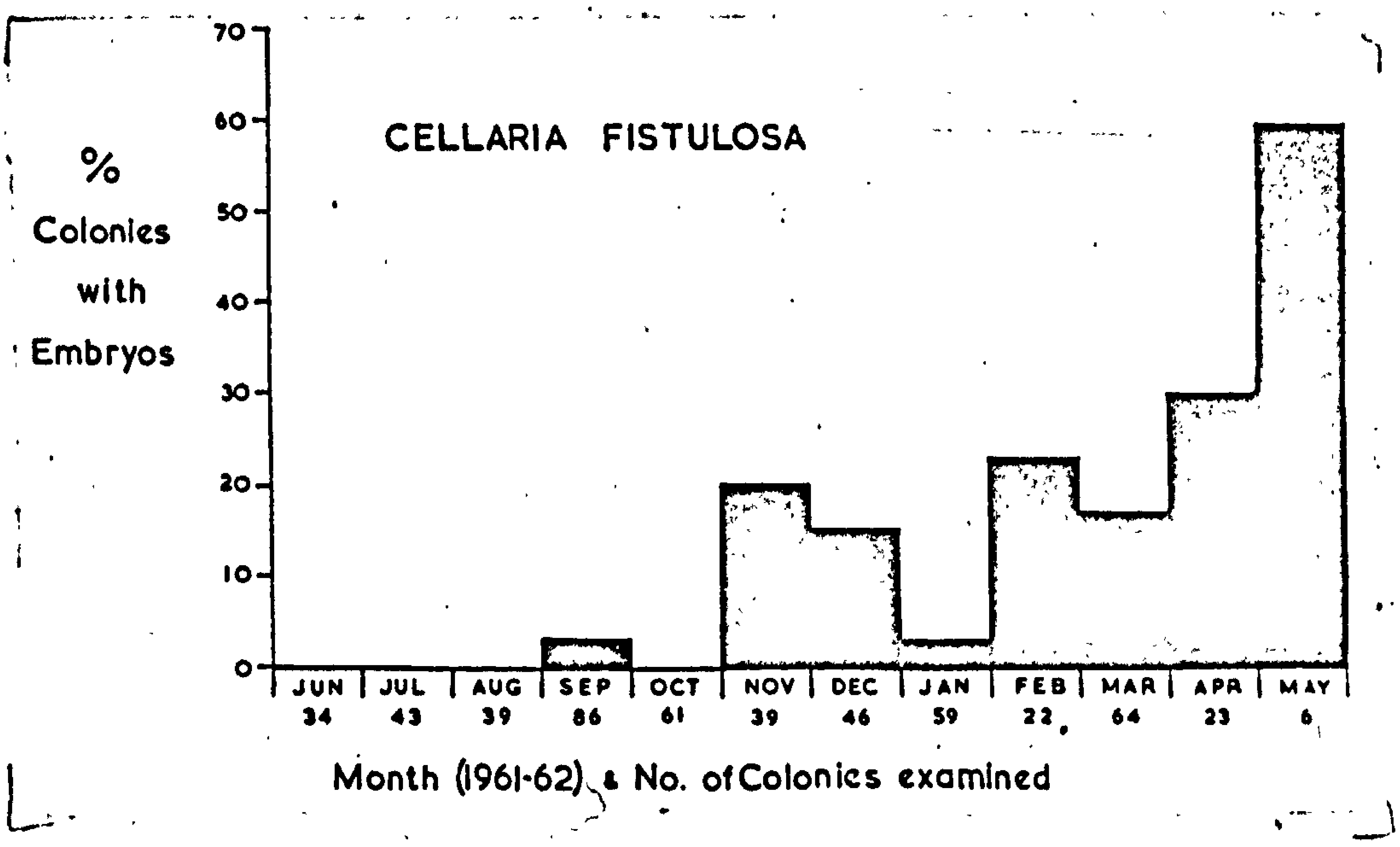


Fig. II. 21. Cellaria fistulosa. Percentage colonies with embryos in each months samples from June 1961 to May 1962.

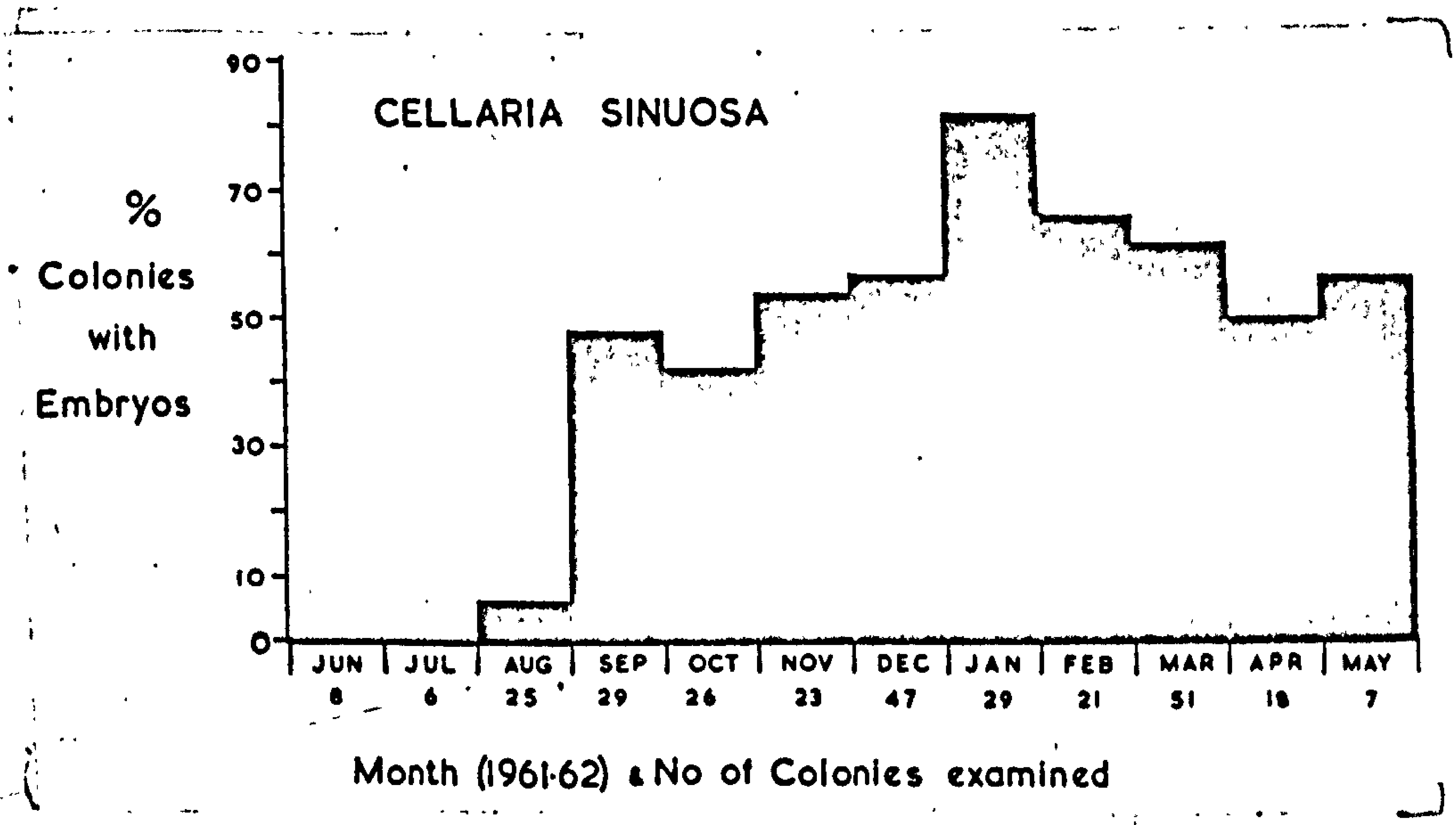


Fig. II. 22. Cellaria sinuosa. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

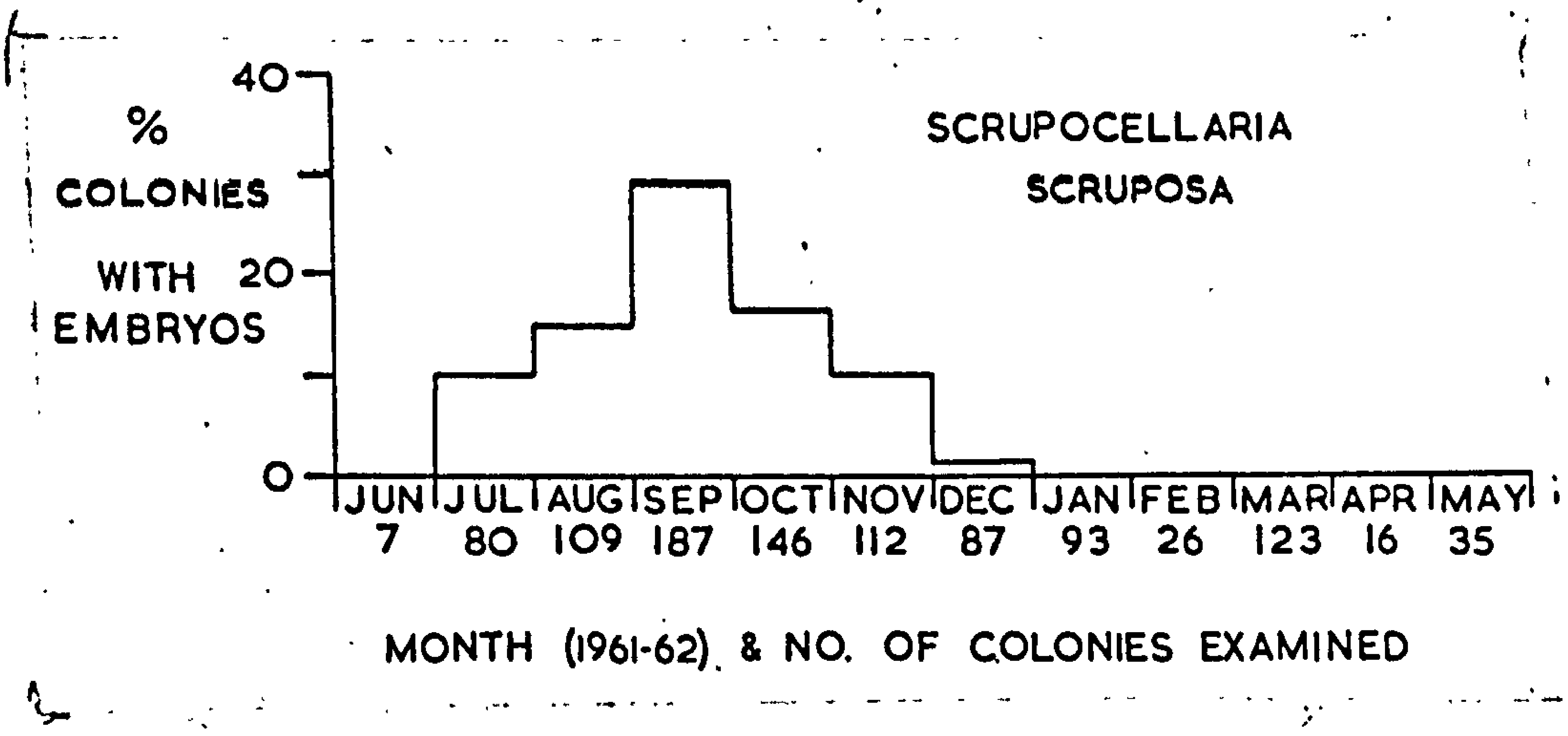


Fig. II. 23. Scrupocellaria scruposa. Percentage colonies with embryos in each month's samples from June1961 to May 1962

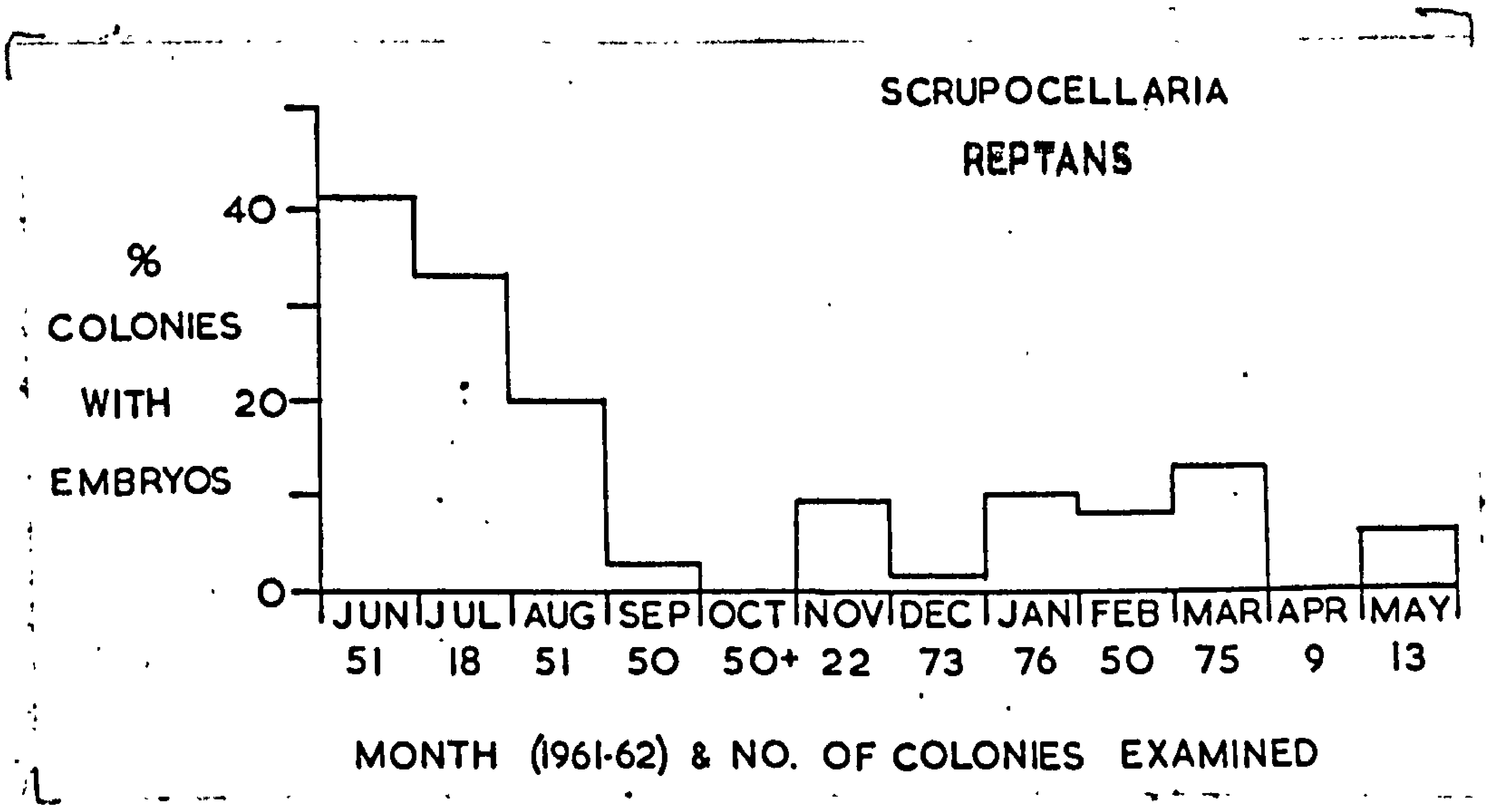


Fig. II.24. Scrupocellaria reptans. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

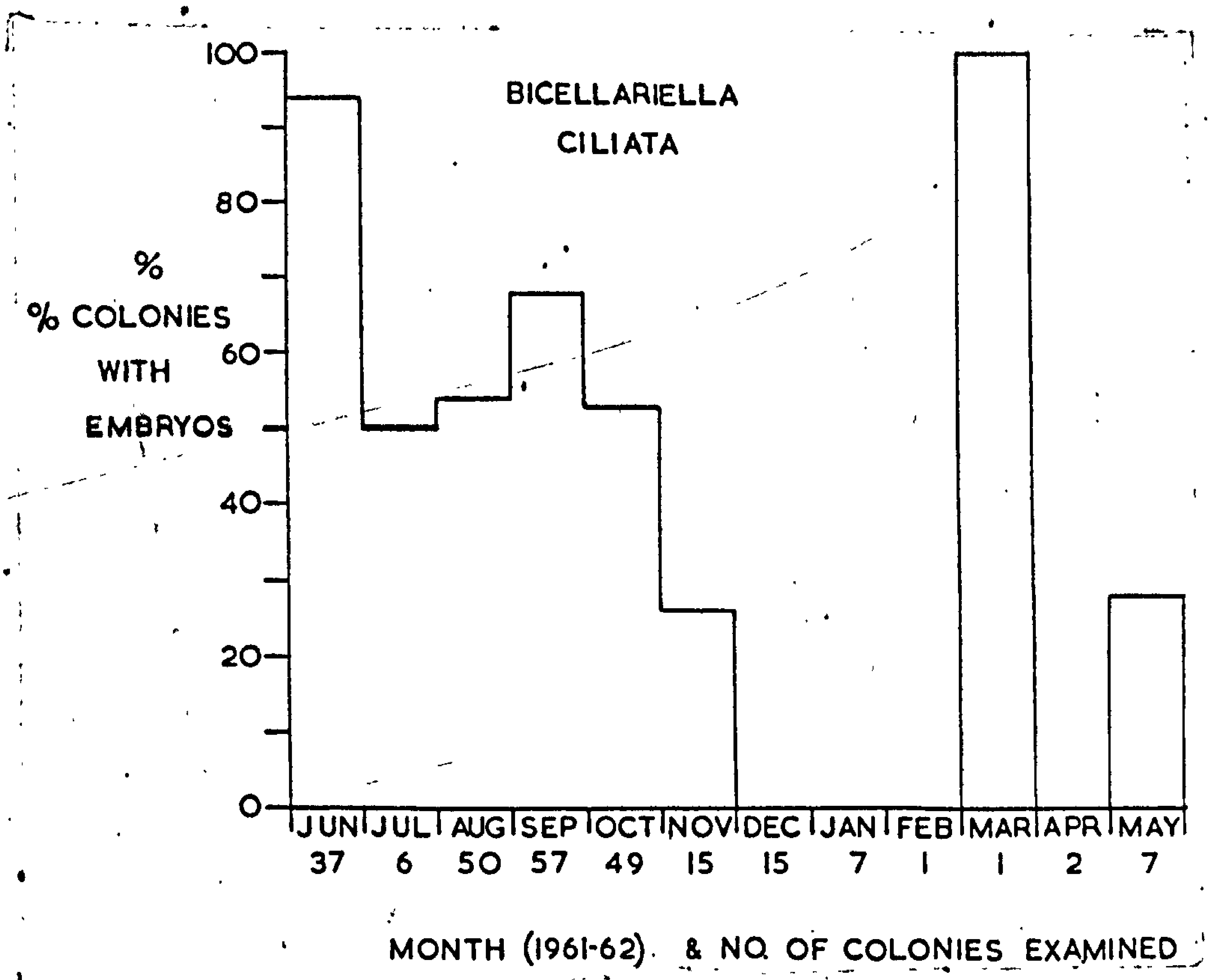


Fig. II. 25. Bicellariella ciliata. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

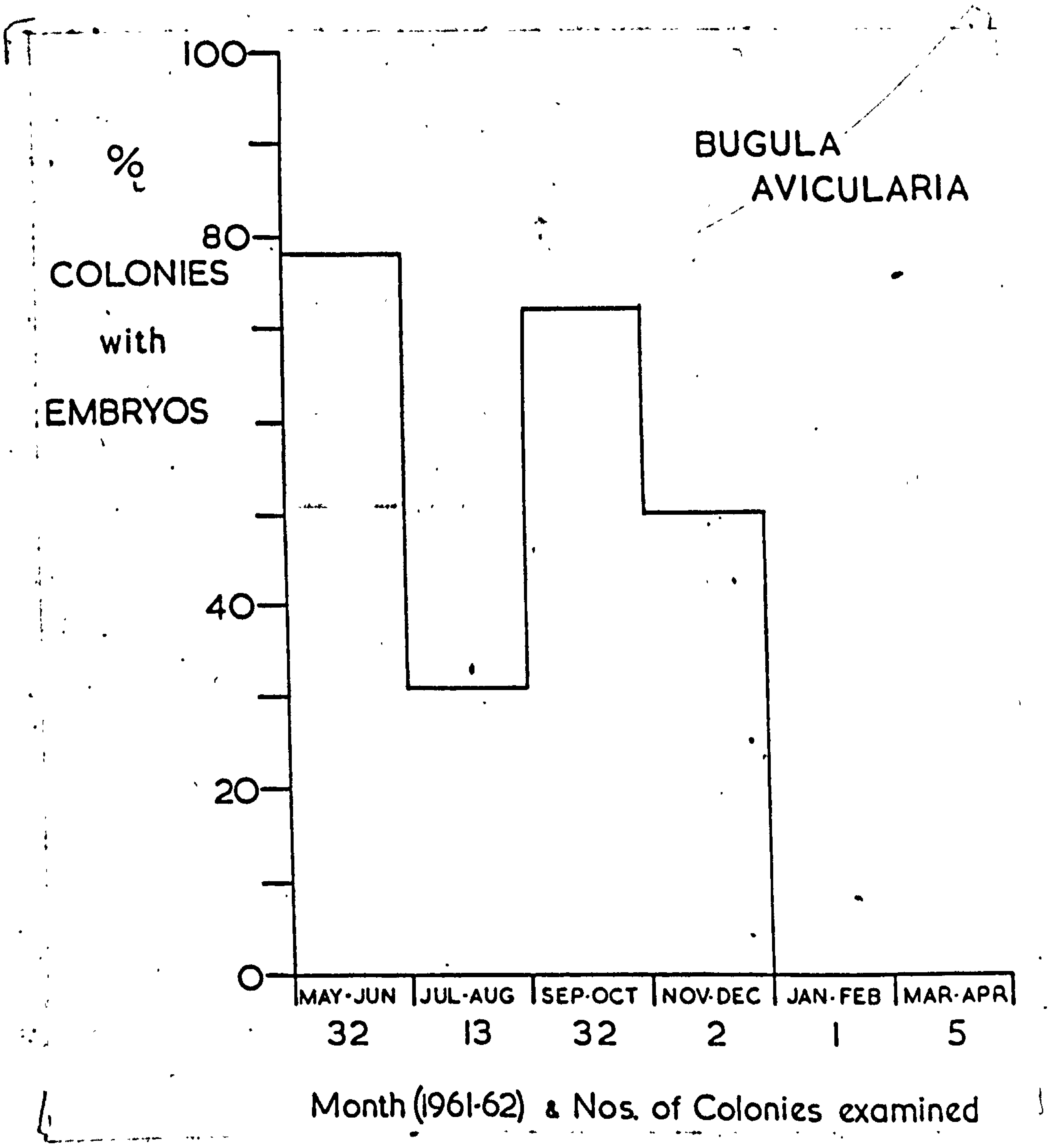


Fig. II. 26. Bugula avicularia. Percentage colonies with embryos in each two month's samples from May 1961 to April 1962.

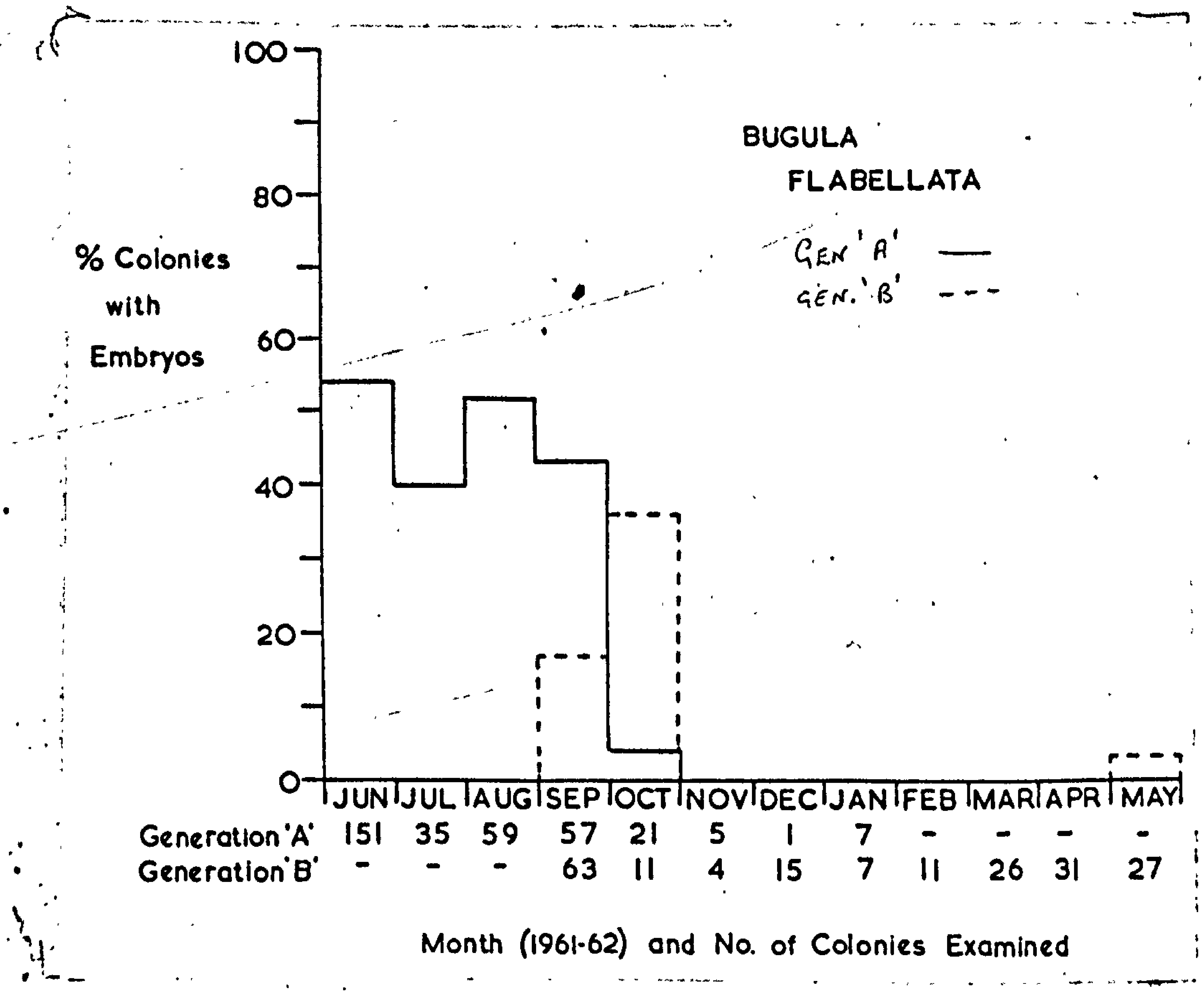


Fig. II. 27. Bugula flabellata. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

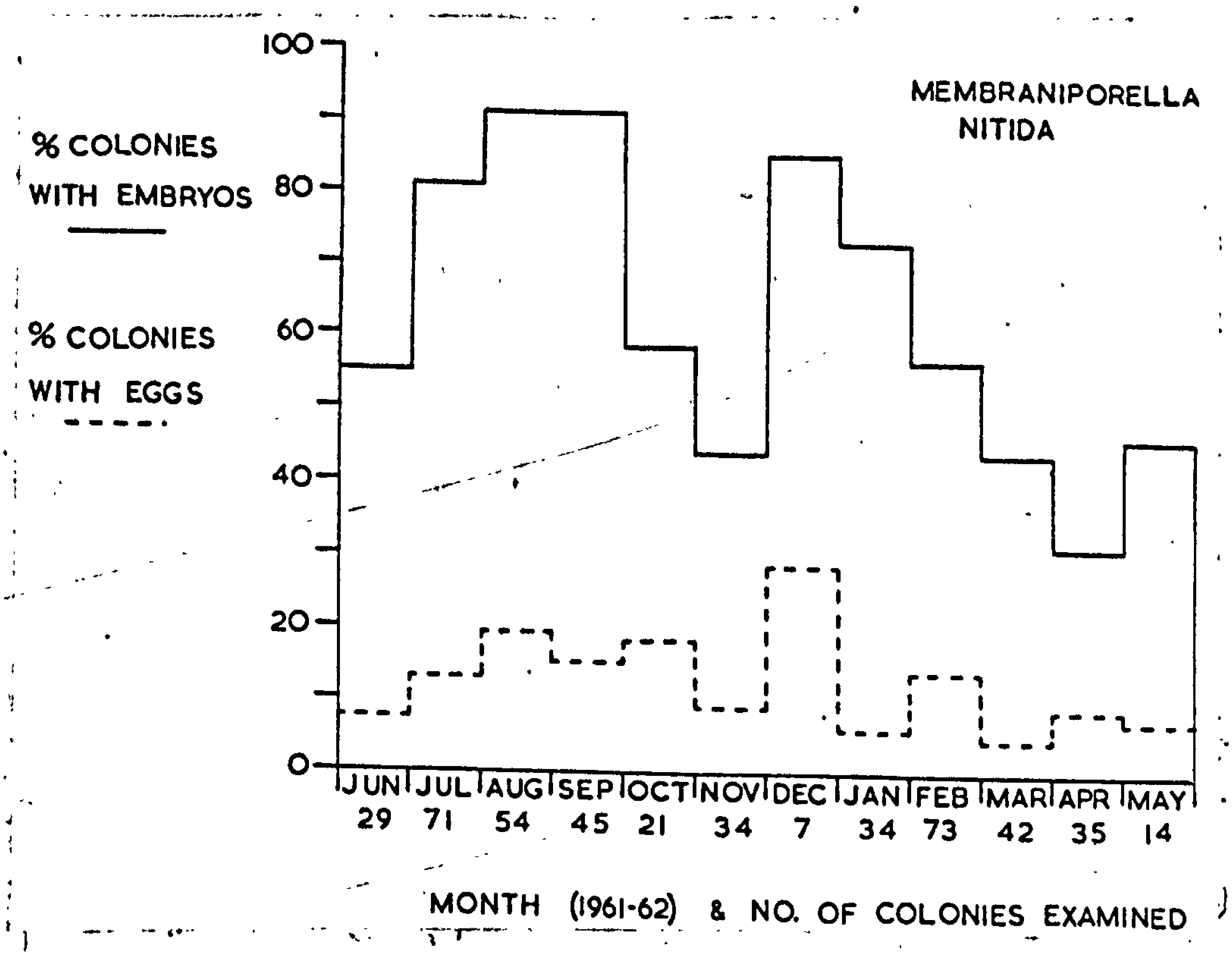


Fig. II.28. Membraniporella nitida. Percentages of colonies with embryos and with eggs in each month's samples from June 1961 to May 1962.

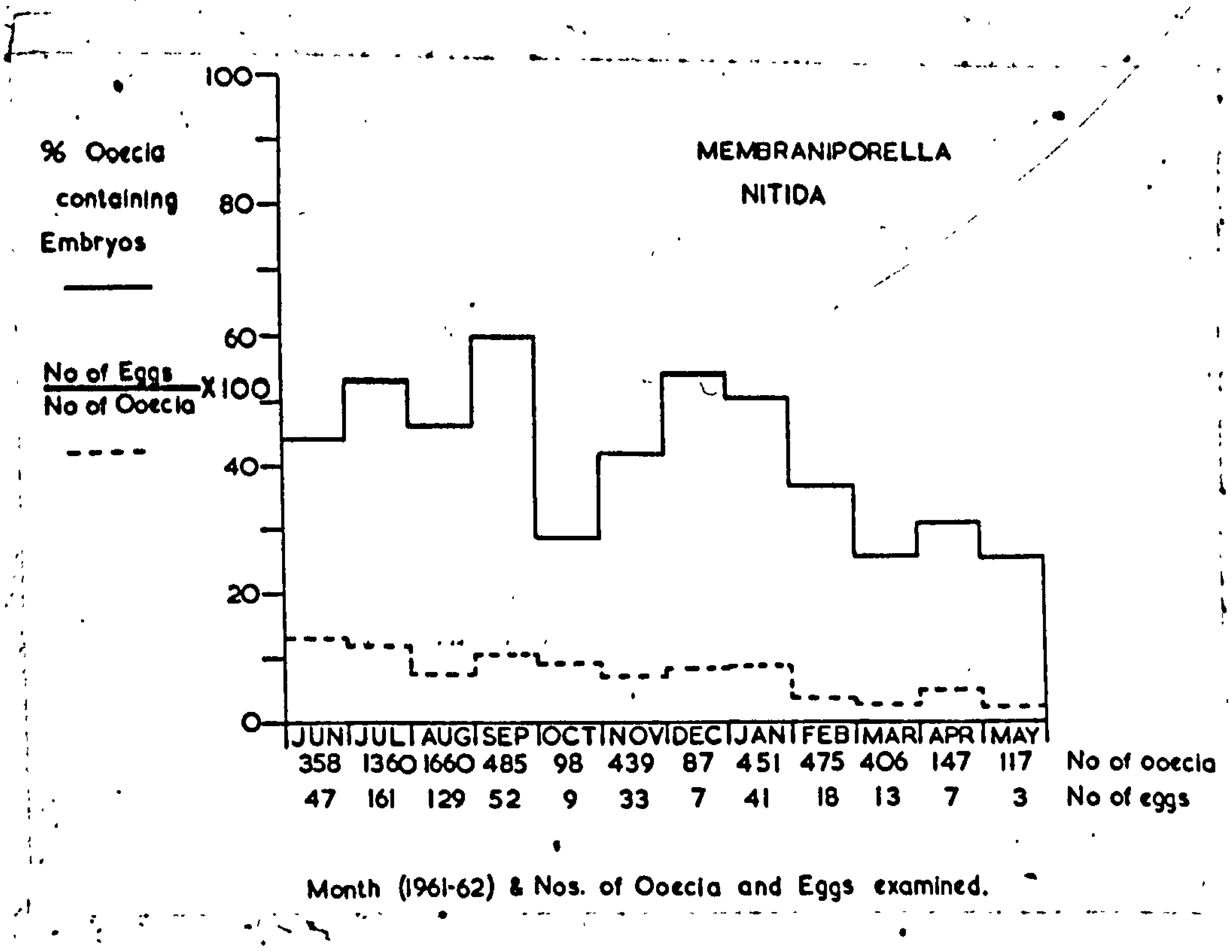


Fig. II.29. Membraniporella nitida. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia) x 100 for each month's samples from June 1961 to May 1962.

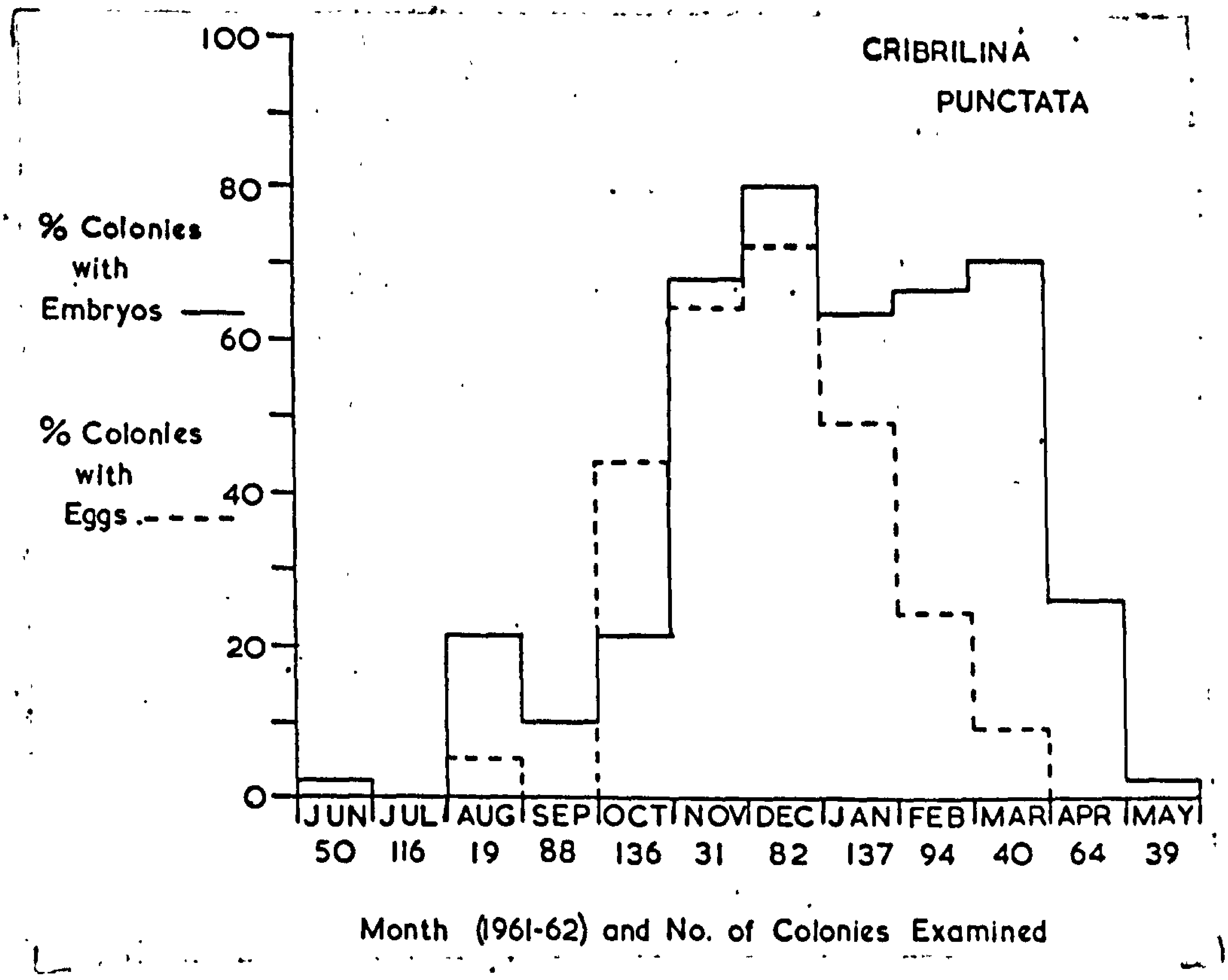


Fig. II.30. Cribrilina punctata. Percentages of colonies with embryos and with eggs in each Month's samples from June 1961 to May 1962.

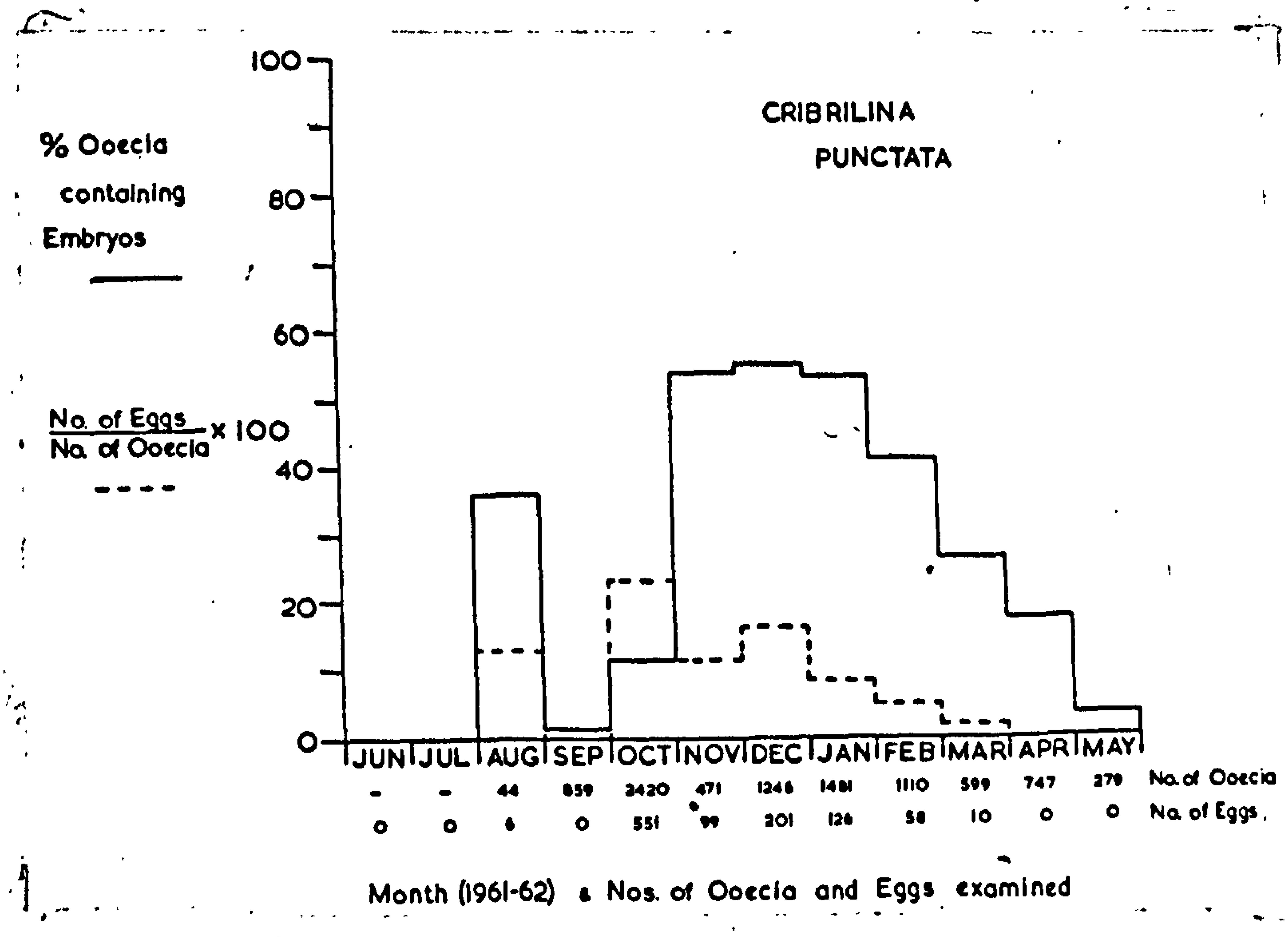


Fig.II.31. Cribrilina punctata. Percentage ooecia containing embryos and (no. of eggs: no. of ooecia)x100 for each month's samples from June 1961 to May 1962.

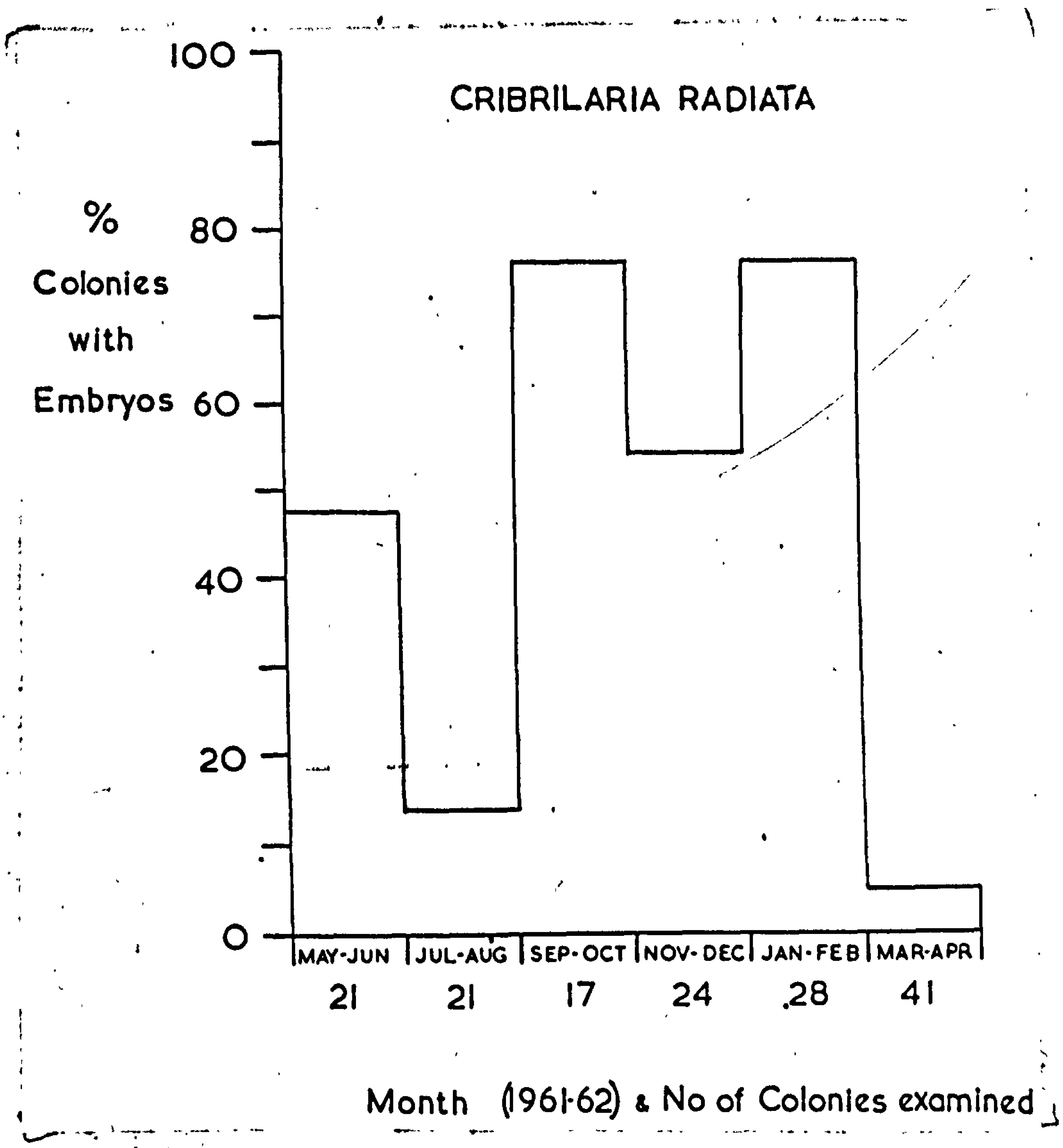


Fig. II. 32. Cribrilaria radiata. Percentage colonies with embryos in each two month's samples from May 1961 to April 1962.

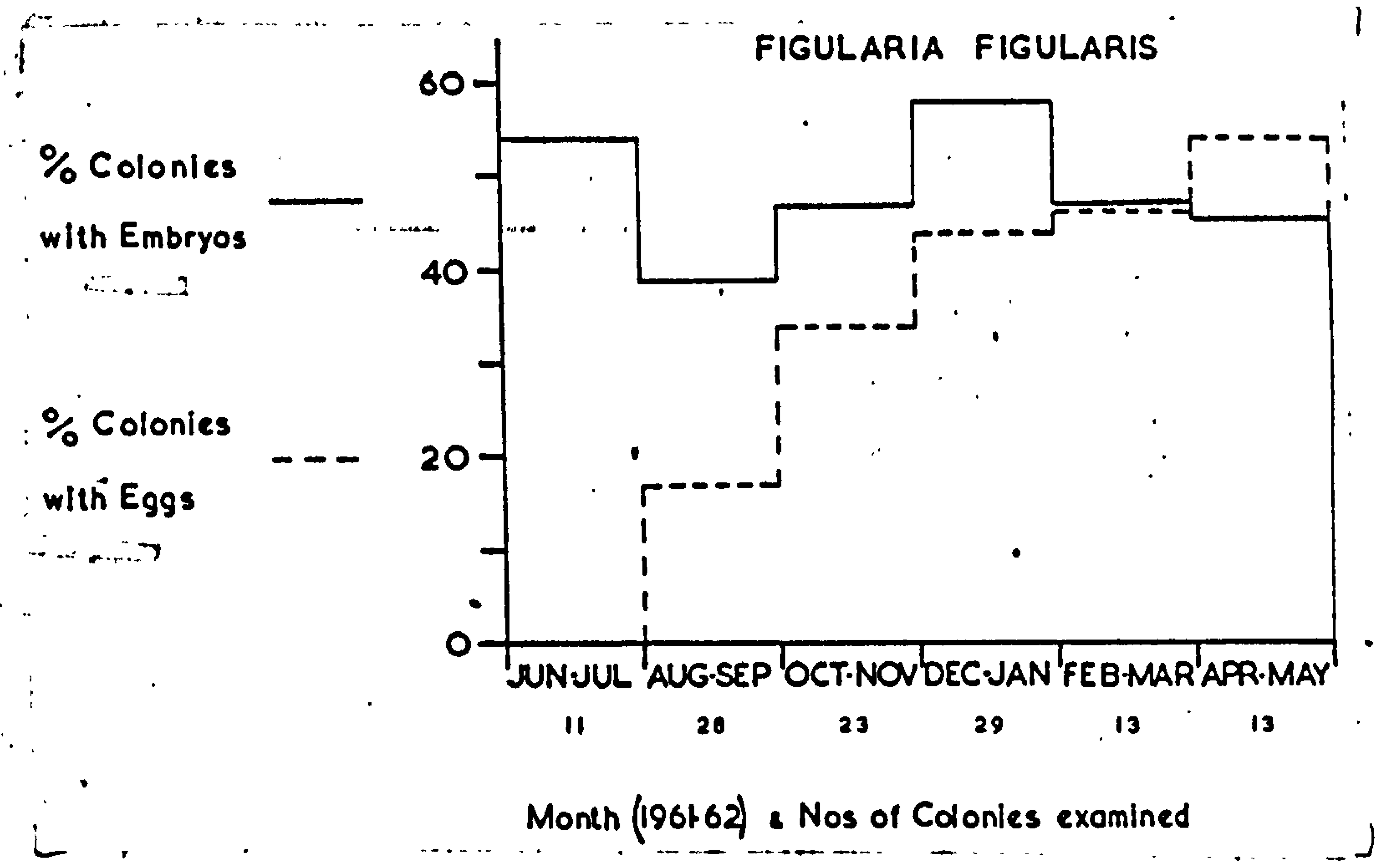


Fig. II.33. Figularia figularis. Percentages of colonies with embryos and percentages with eggs in each two month's samples from June 1961 to May 1962.

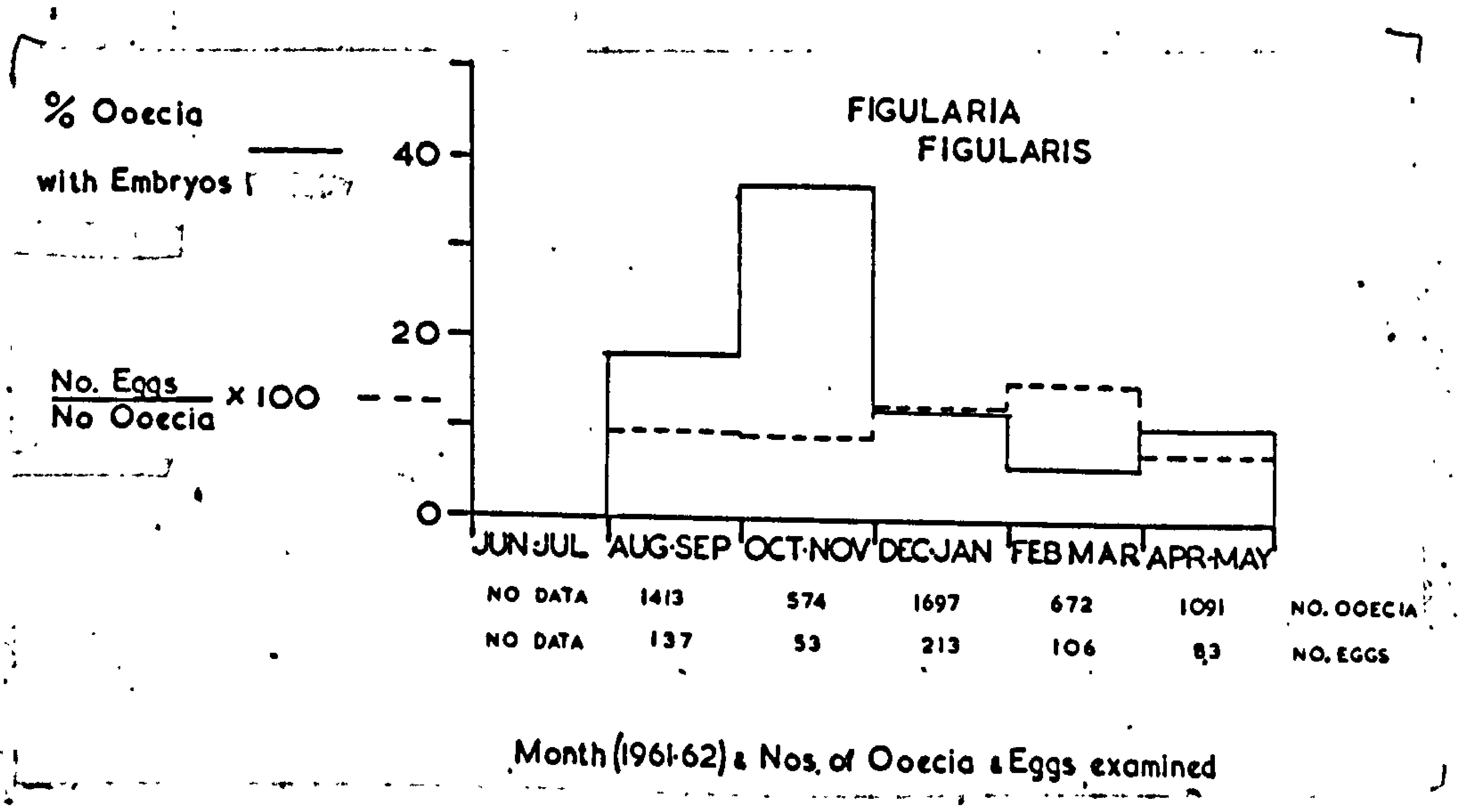


Fig II. 34. Figularia figularis. Pecentages of ooecia containing embryos and (No. of eggs; no. of ooecia)100 for each two month's samples from June 1961 to May 1962.

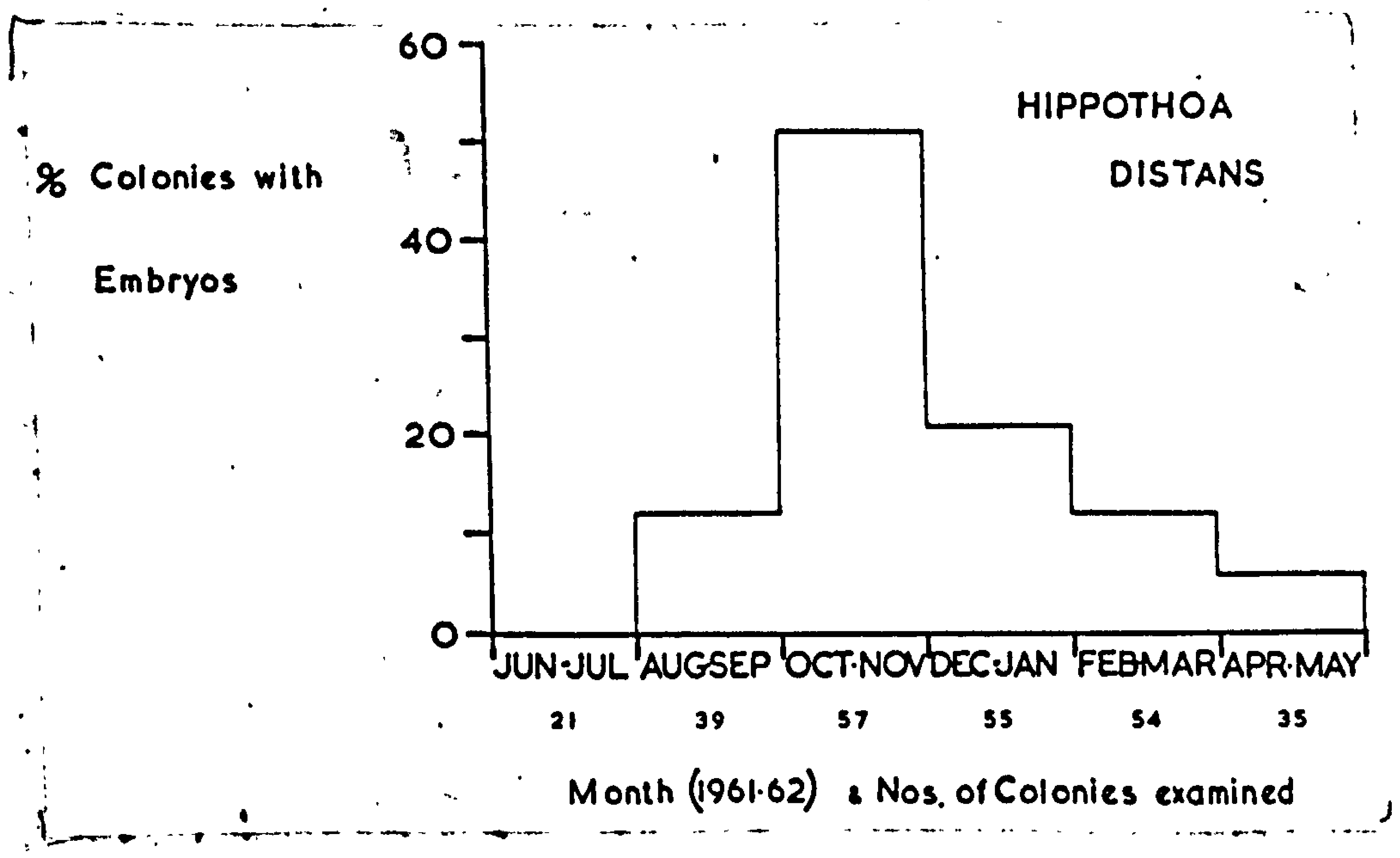


Fig. II.35. Hippothoa distans. Pecentage colonies with embryos in each two month's samples from June 1961 to May 1962.

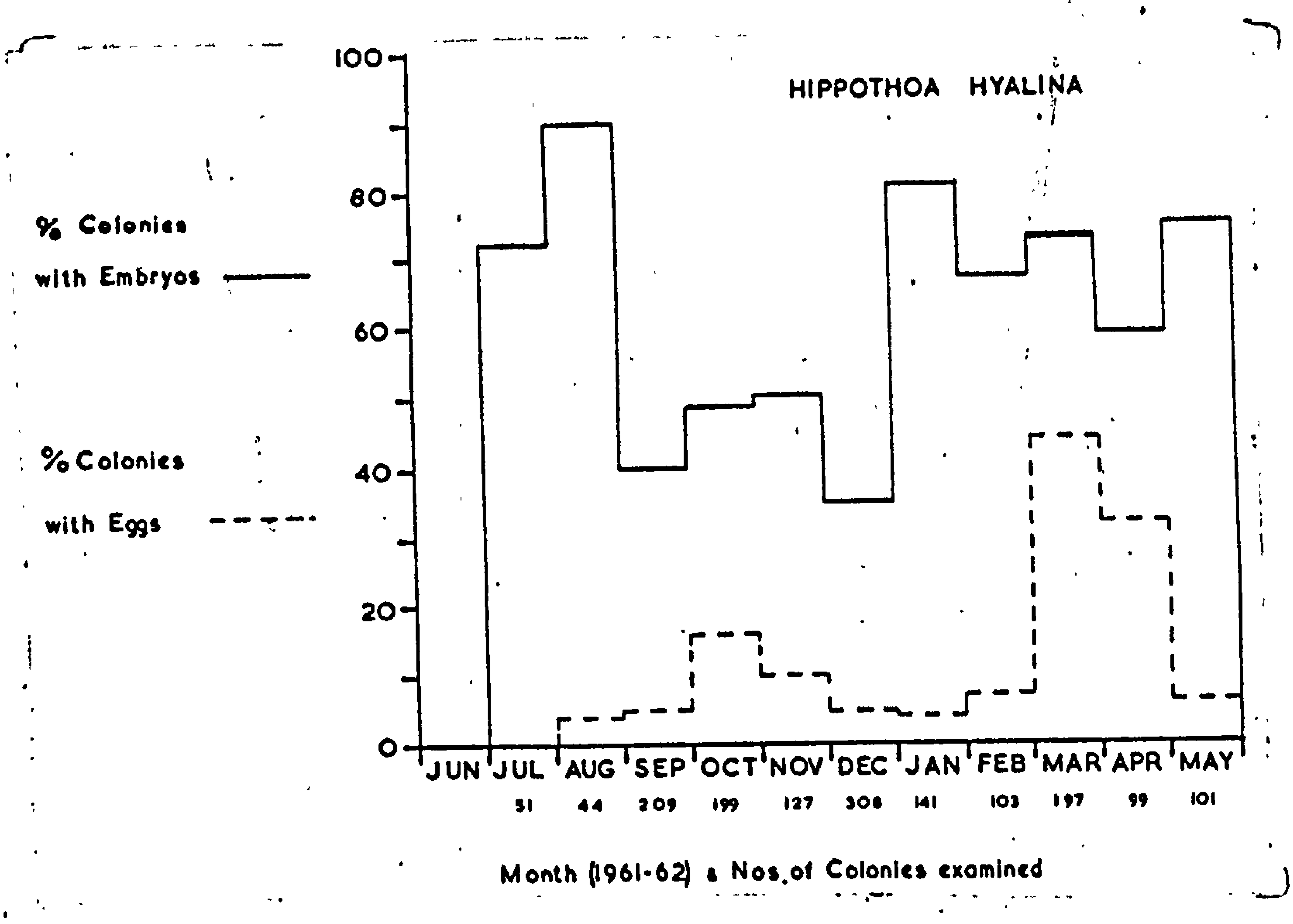


Fig. II. 36. Hippothoa hyalina .Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May 1962.

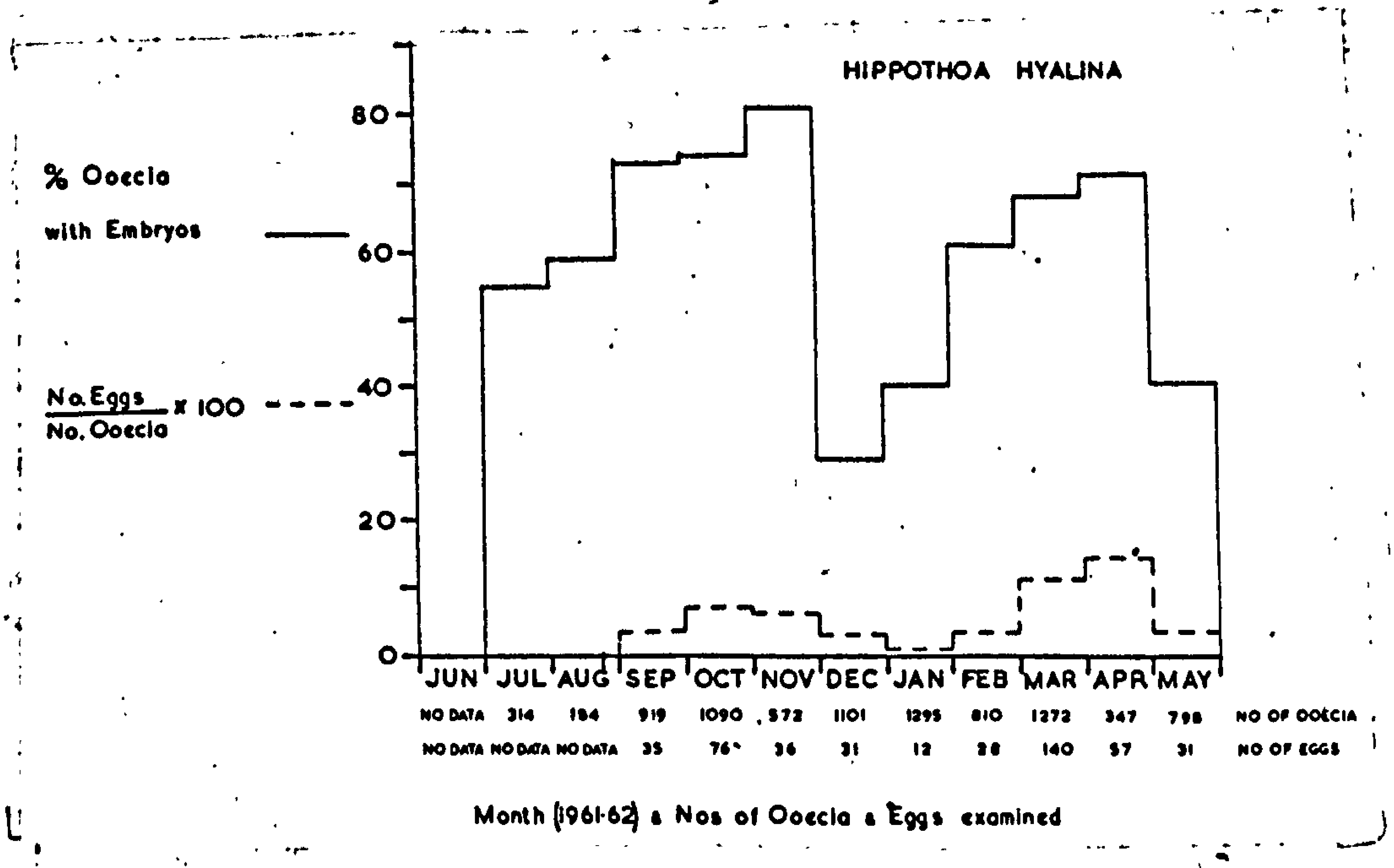


Fig. II. 37. Hippothoa hyalina. Percentage ooecia containing embryos and (No. of eggs: no. of ooecia)100 for each months samples from June 1961 to May 1962.

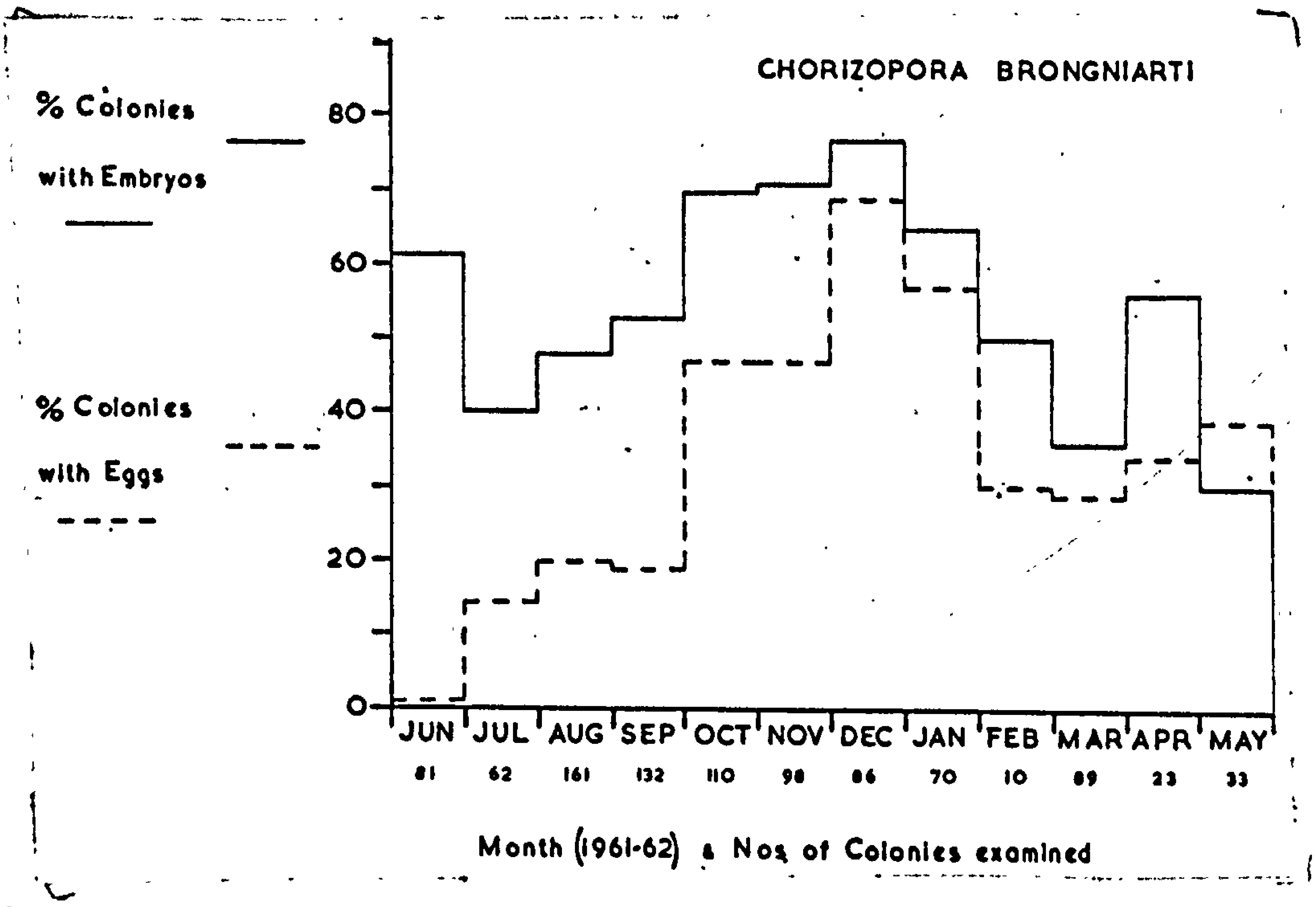


Fig. II. 38. Chorizopora brongniarti. Percentage colonies with embryos and percentage with eggs in each months samples from June 1961 to May 1962.

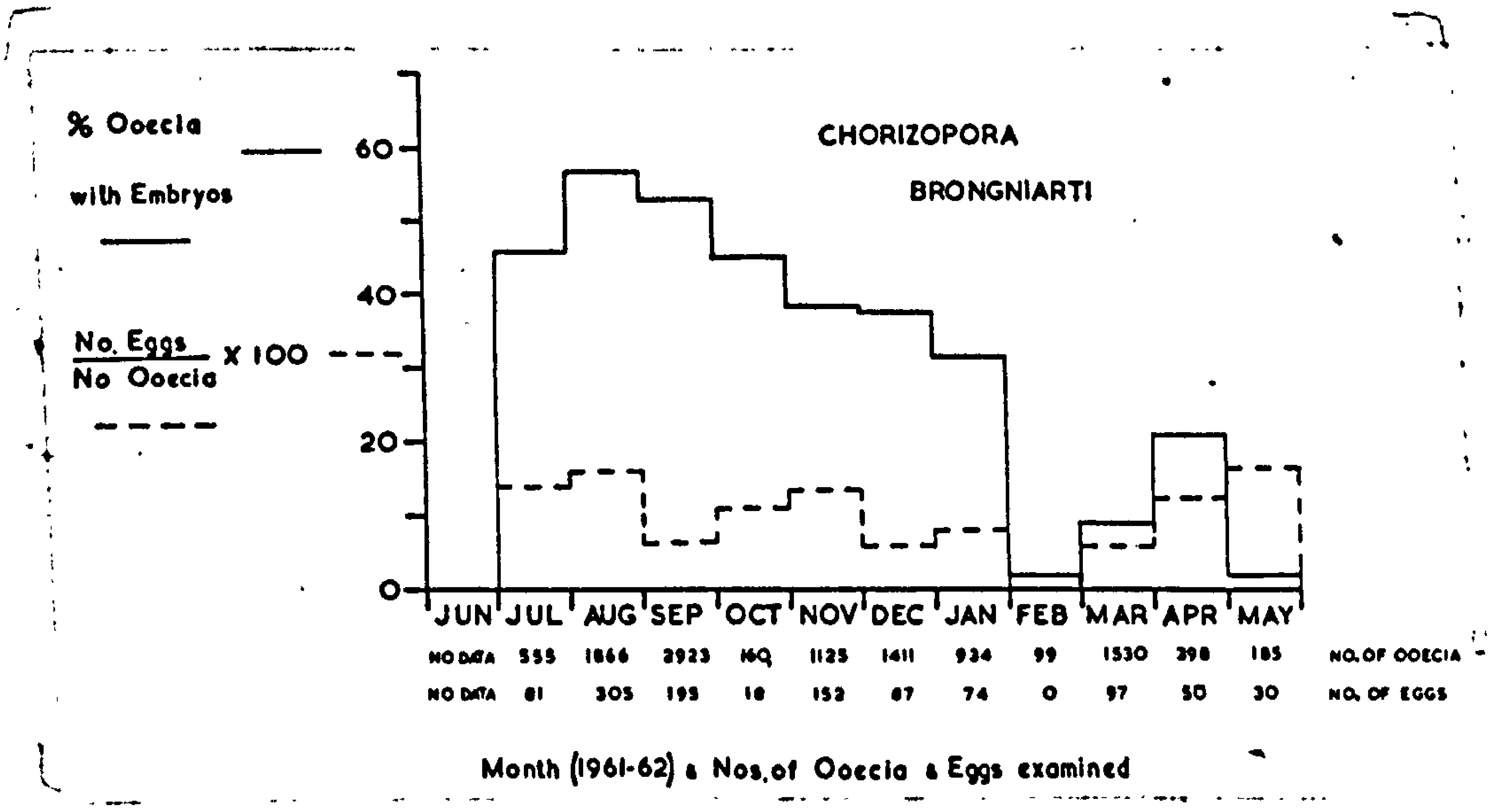


Fig. II. 39. Chorizopora brongniarti. Percentage ooecia containing embryos and (No. of eggs: no. of ooecia)100 for each month's samples from June 1961 to May 1962.

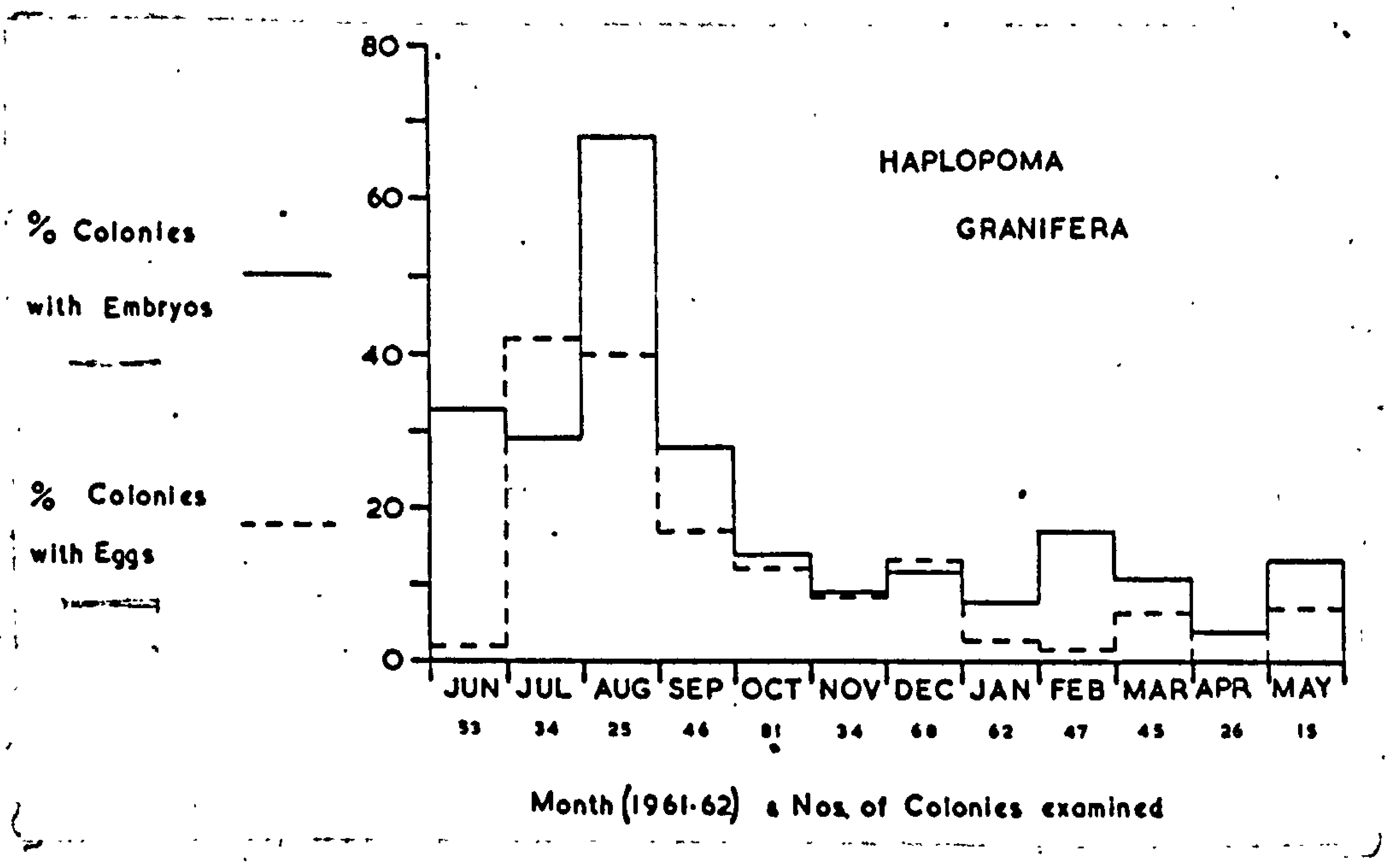


Fig. II.40. Haplopoma graniferum. Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May 1962.

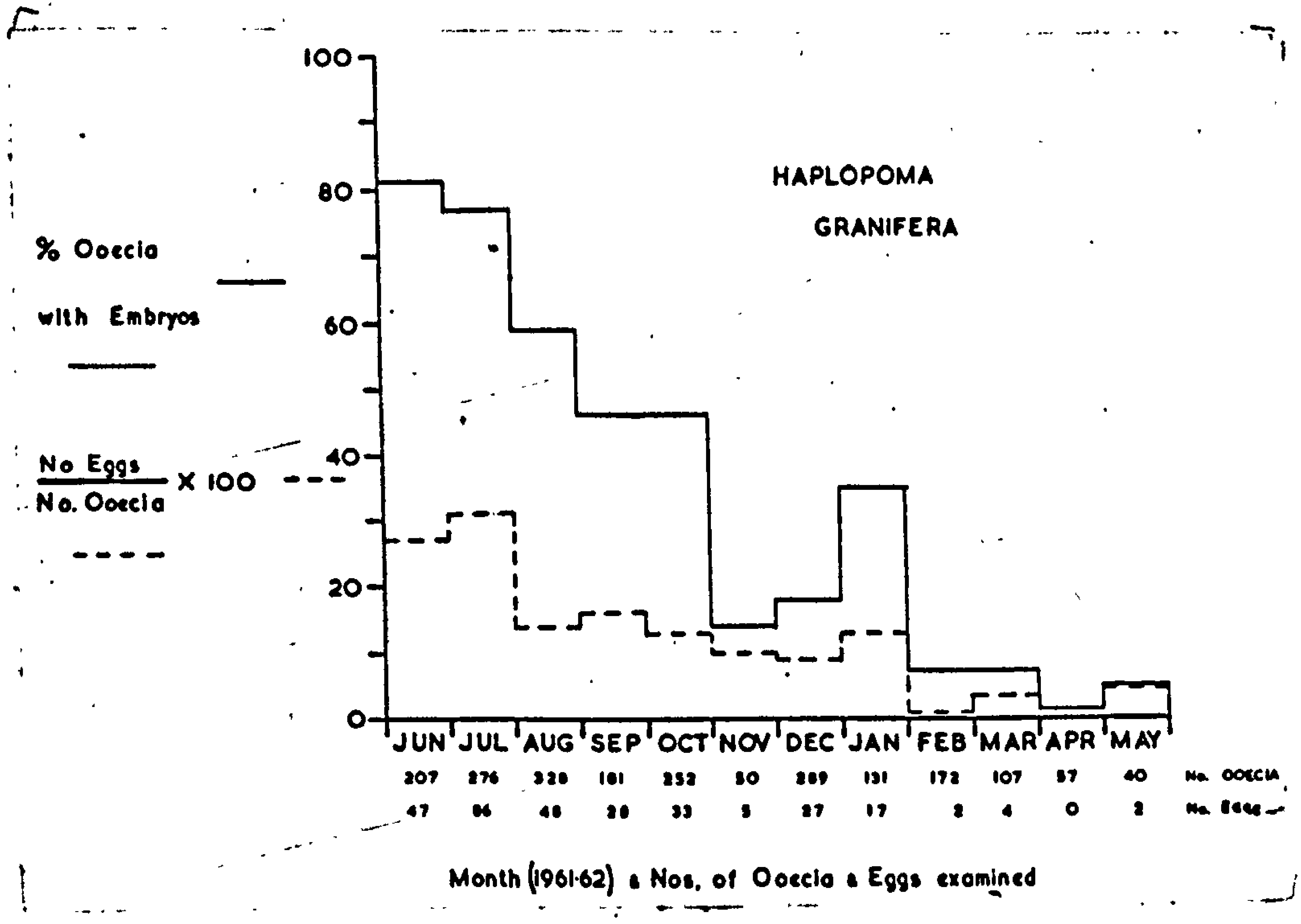


Fig. II. 41. Haplopoma graniferum. Percentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 for each month's samples from June 1961 to May 1962.

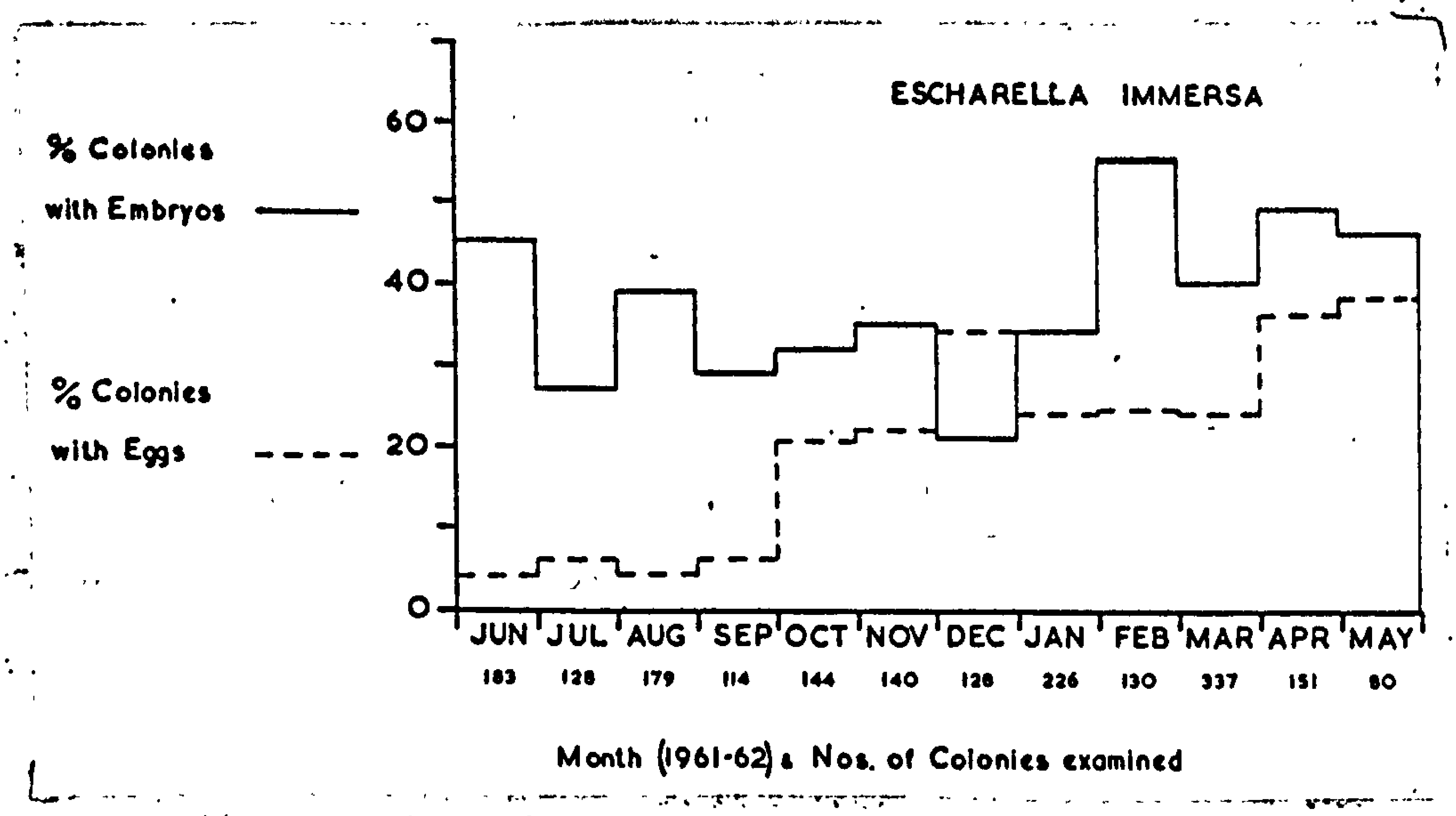


Fig. II. 42. Escharella immersa. Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May1962.

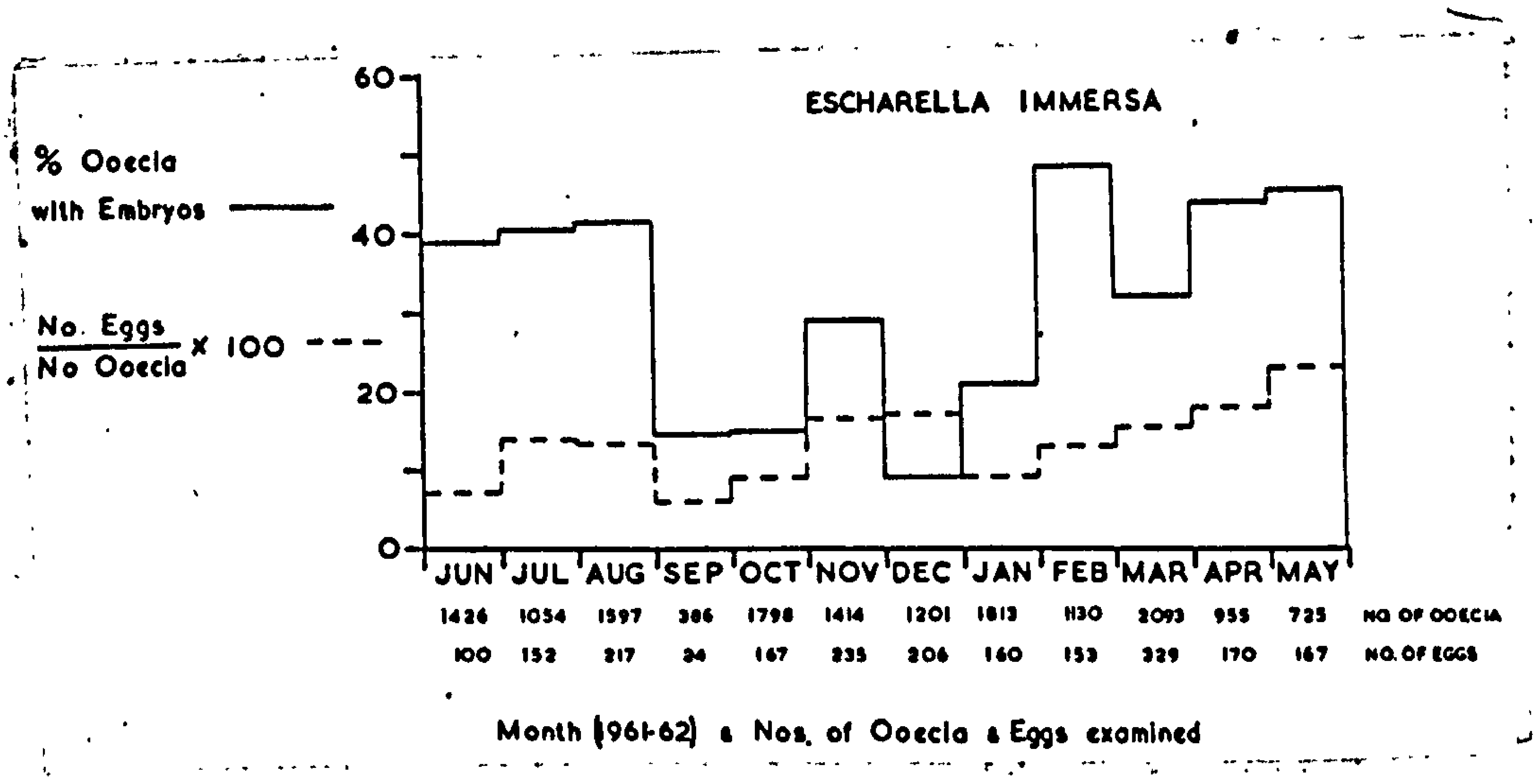


Fig.II. 43. Escharella immersa. Percentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 for each month's samples from June 1961 to May 1962.

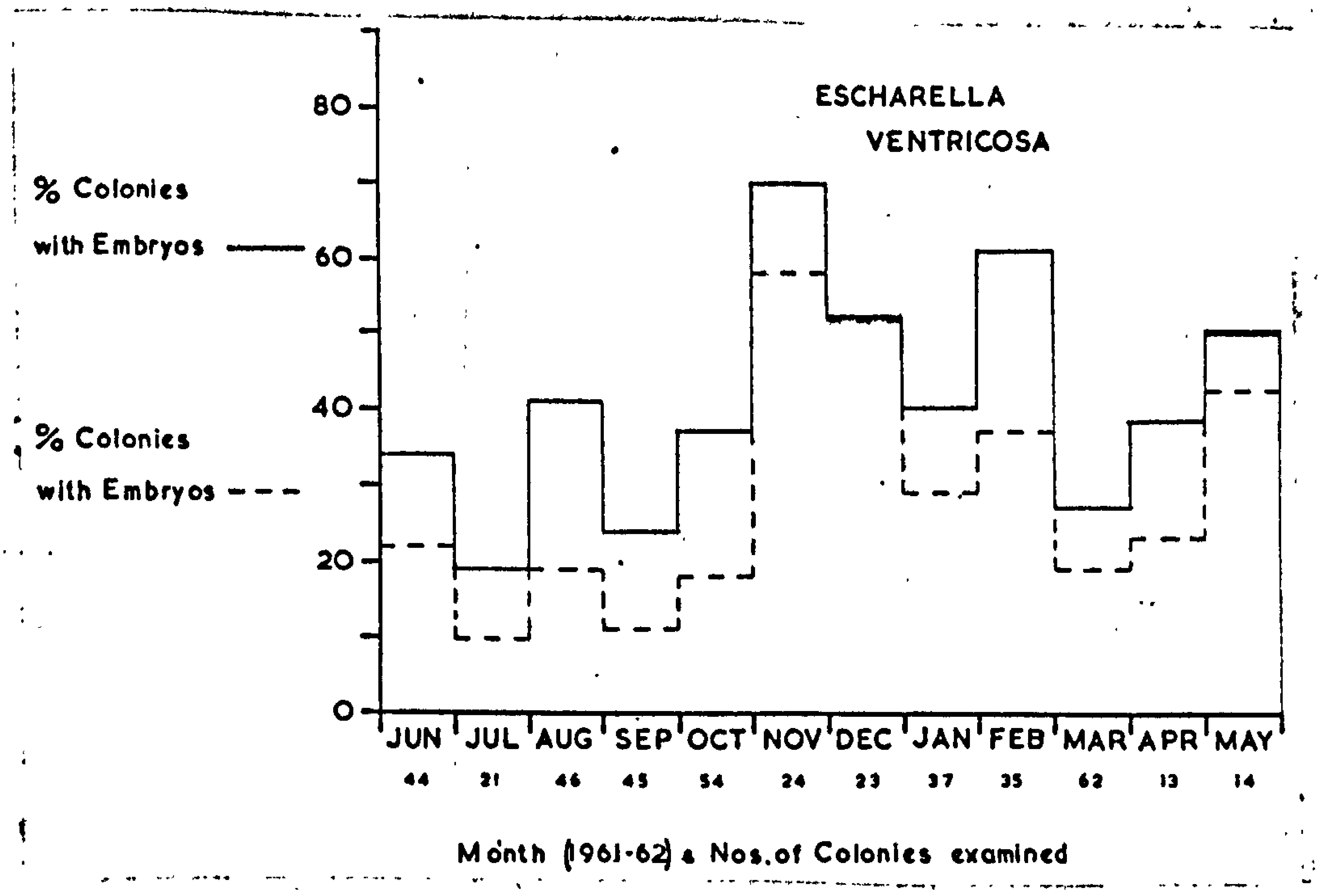


Fig. II. 44. Escharella ventricosa. Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May 1962.

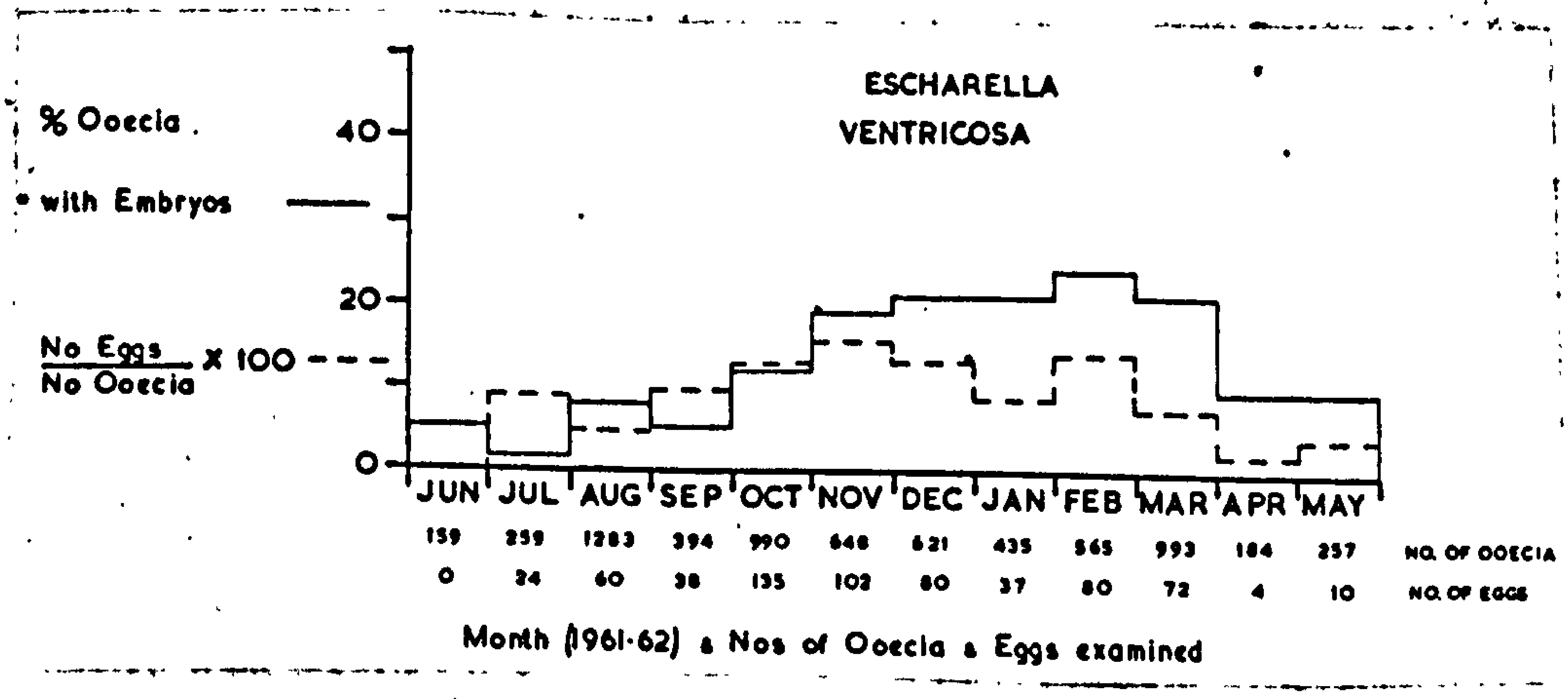


Fig. II. 45. Escharella ventricosa Percentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 for each month's samples from June 1961 to May 1962.

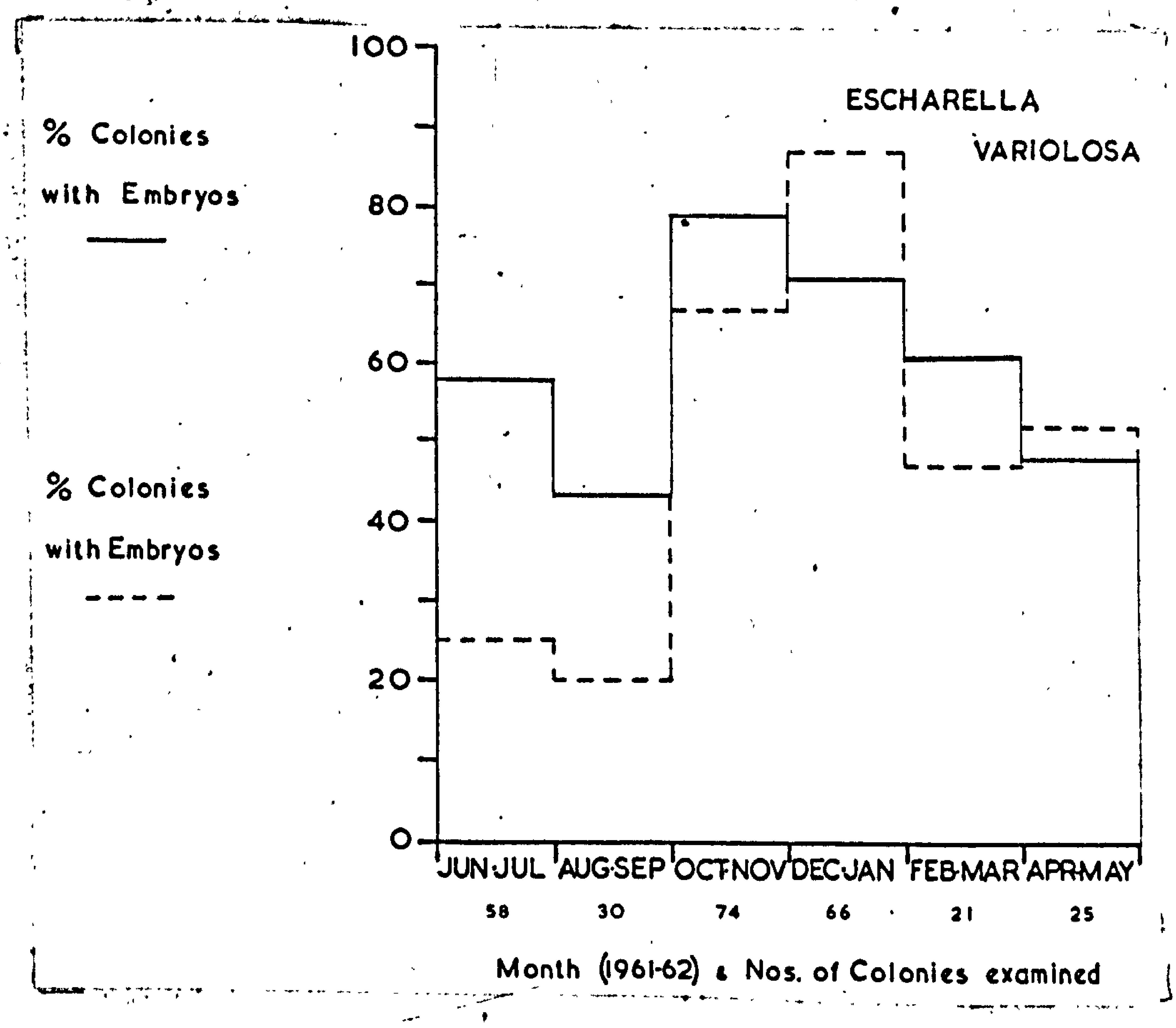


Fig. II. 46. Escharella variolosa. Percentage colonies with eggs and percentage with embryos for each two month's samples from June 1961 to May 1962.

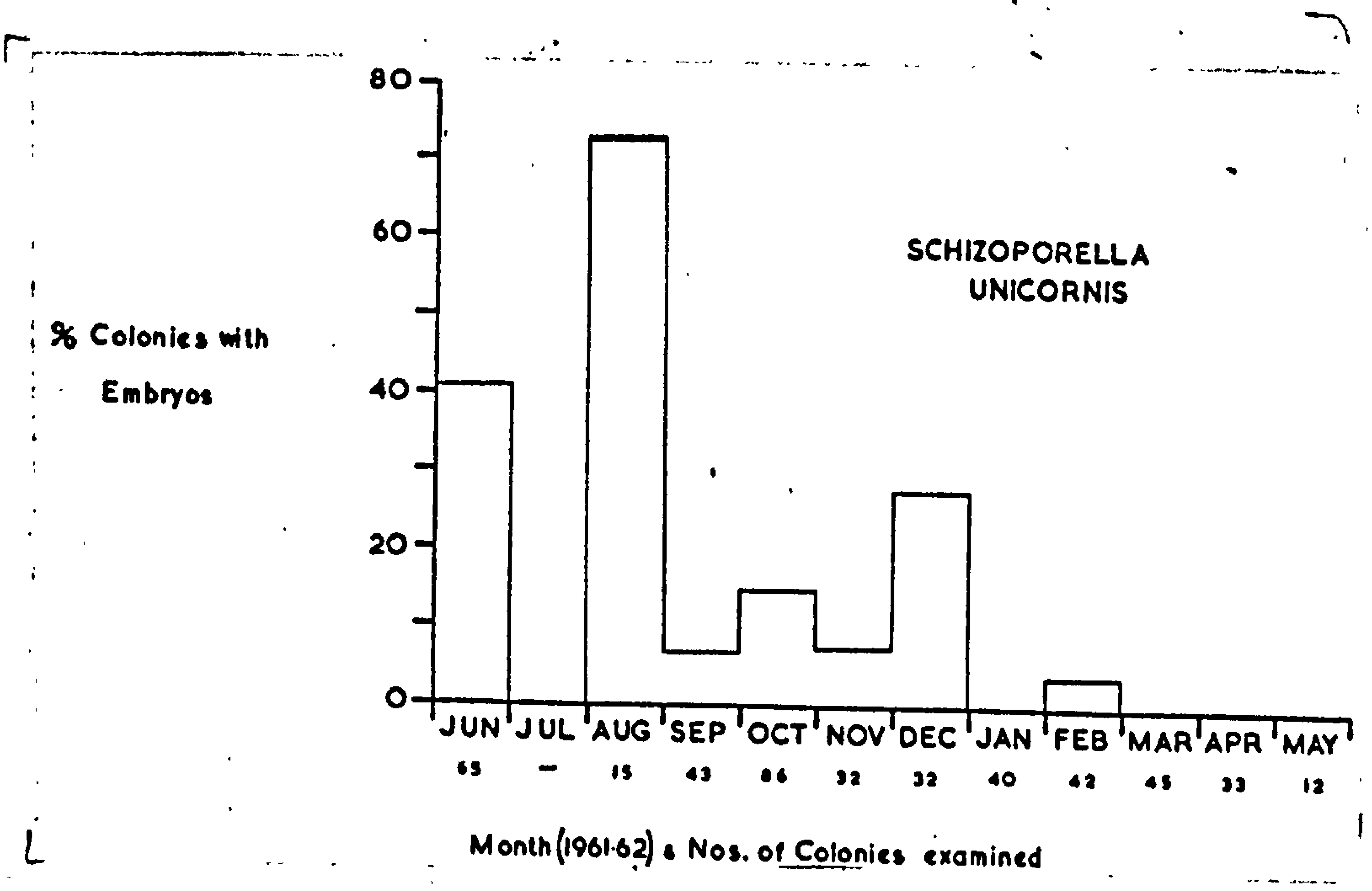


Fig. II.47. <u>Schizoporella unicornis.</u> Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

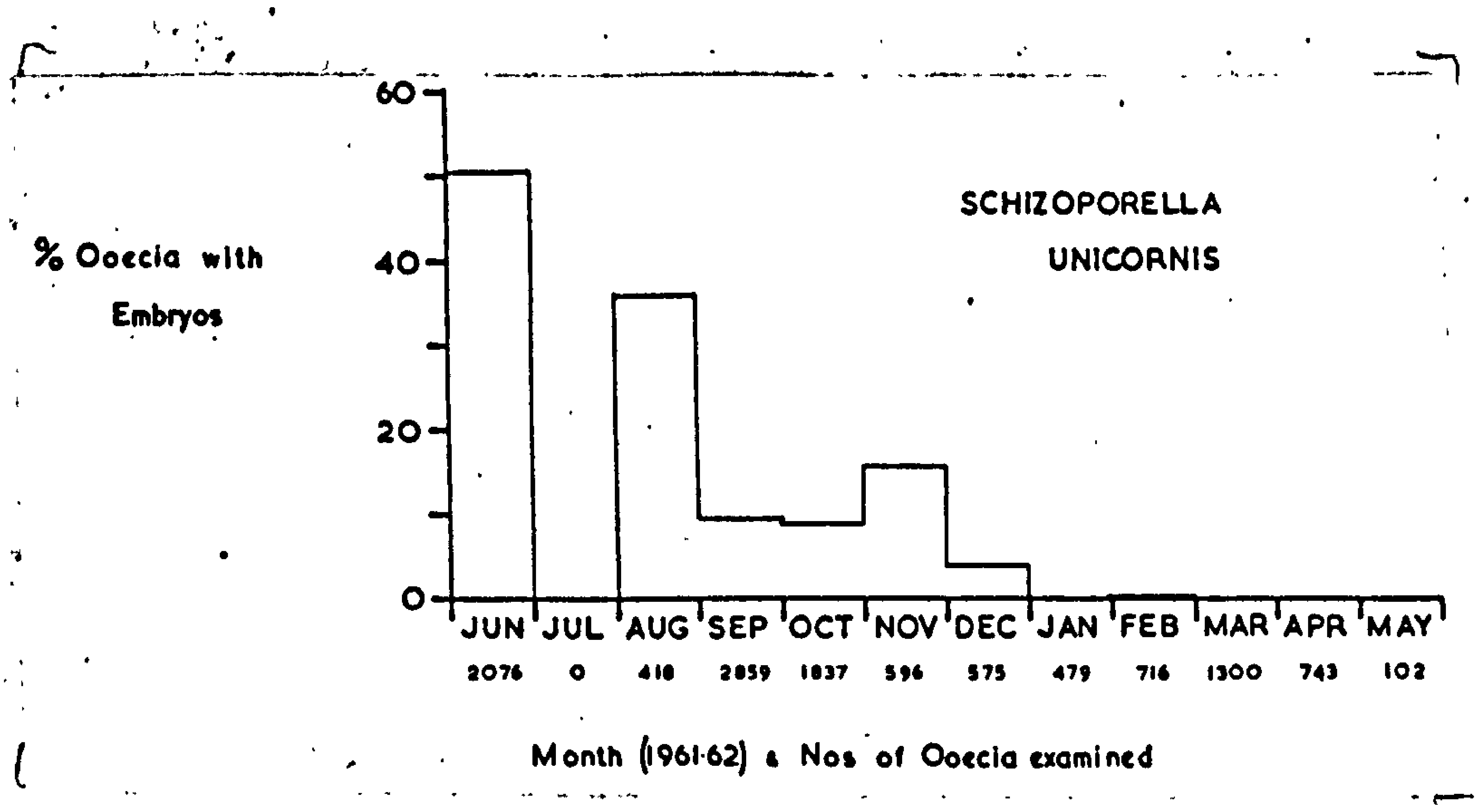


Fig. II.48. <u>Schizoporella unicornis</u>. Percentage ooecia containing embryos in each months samples from June 1961 to May 1962.

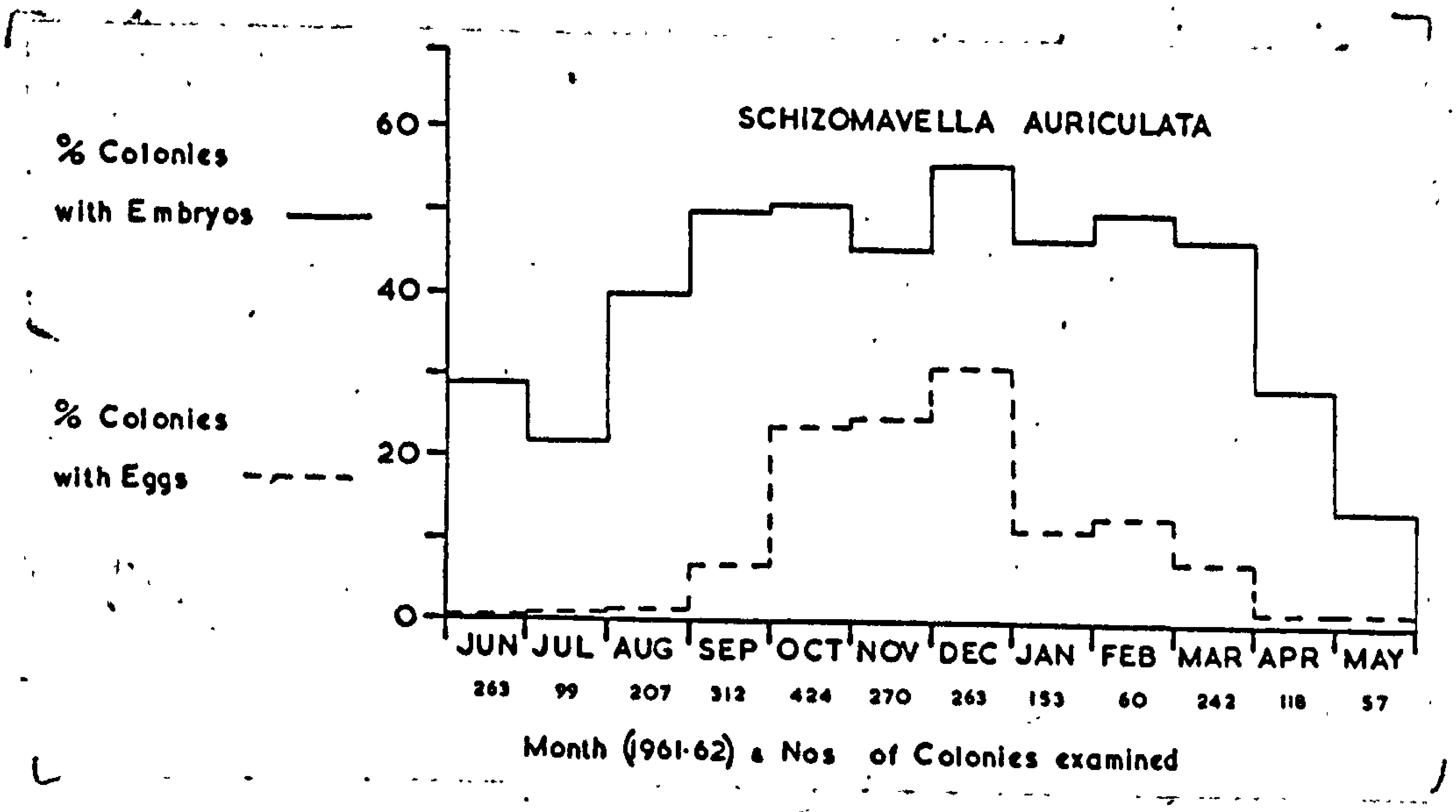


Fig. II.49. Schizomavella auriculata. Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May 1962.

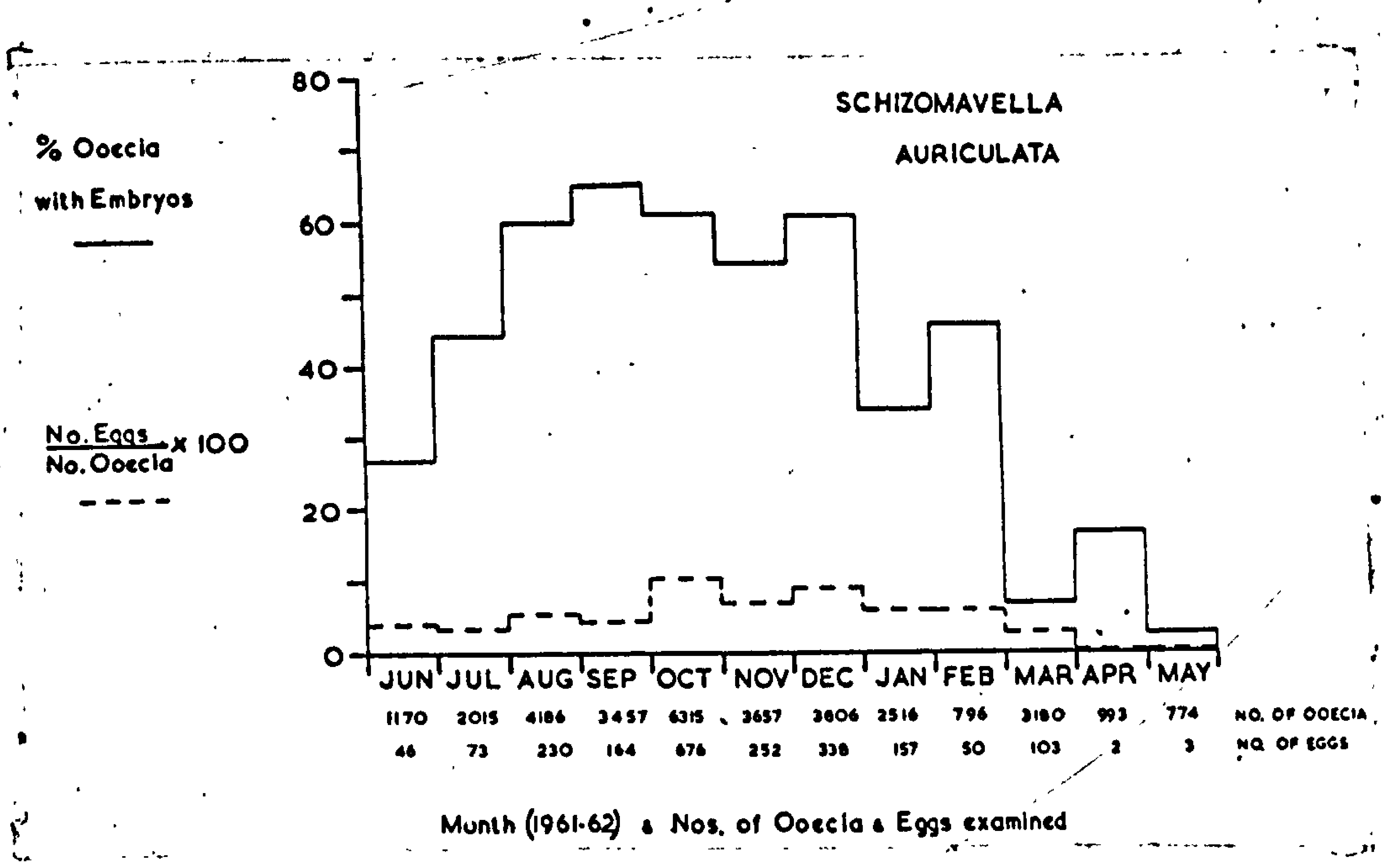


Fig. II.50. Schizomavella auriculata. Pecentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 in each month's samples from June 1961 to May 1962.

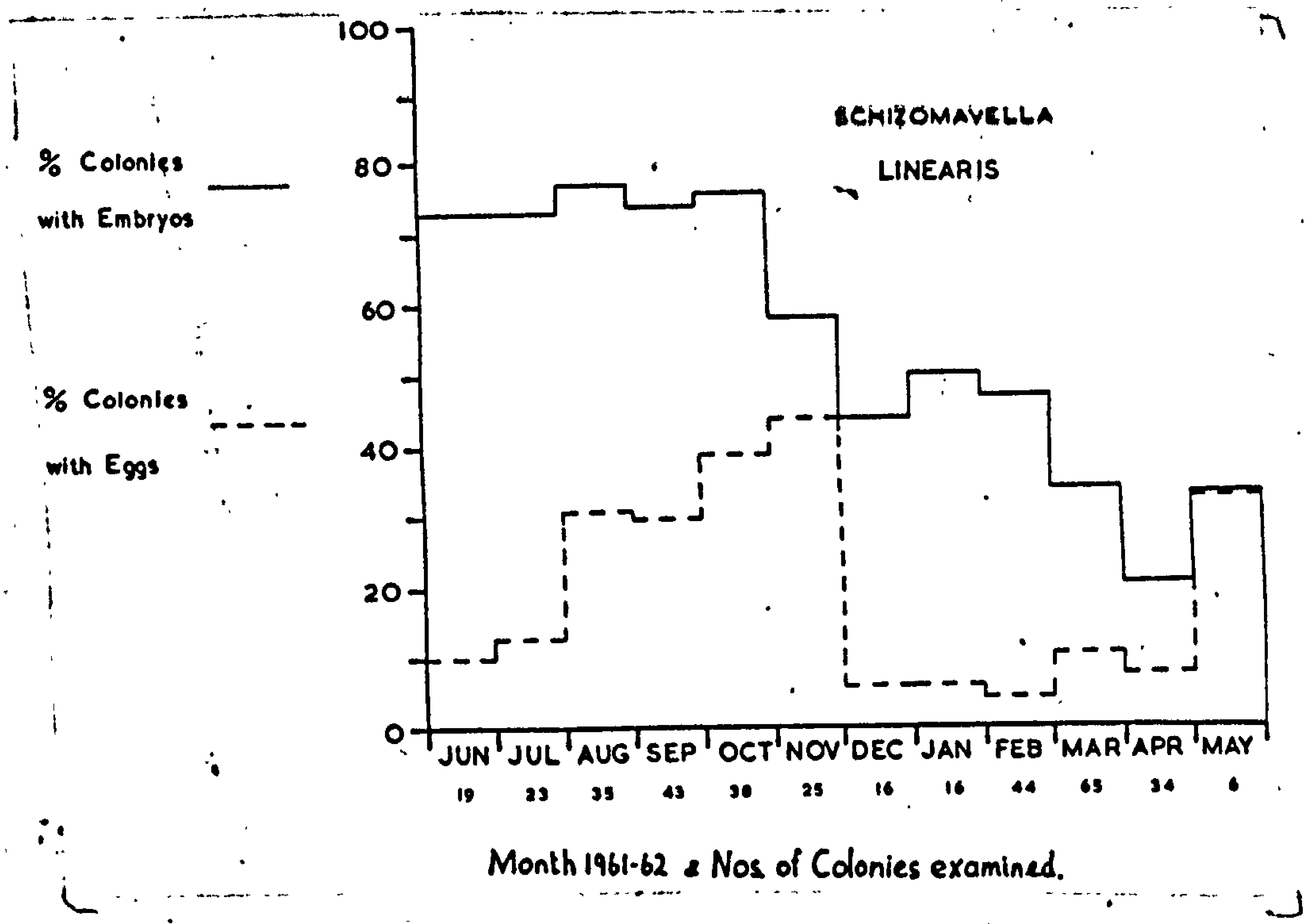


Fig.II. 51. <u>Schizomavella</u> <u>linearis</u>. Pecentage colonies with embryos and percentage with eggs in each months samples from June 1961 to May 1962

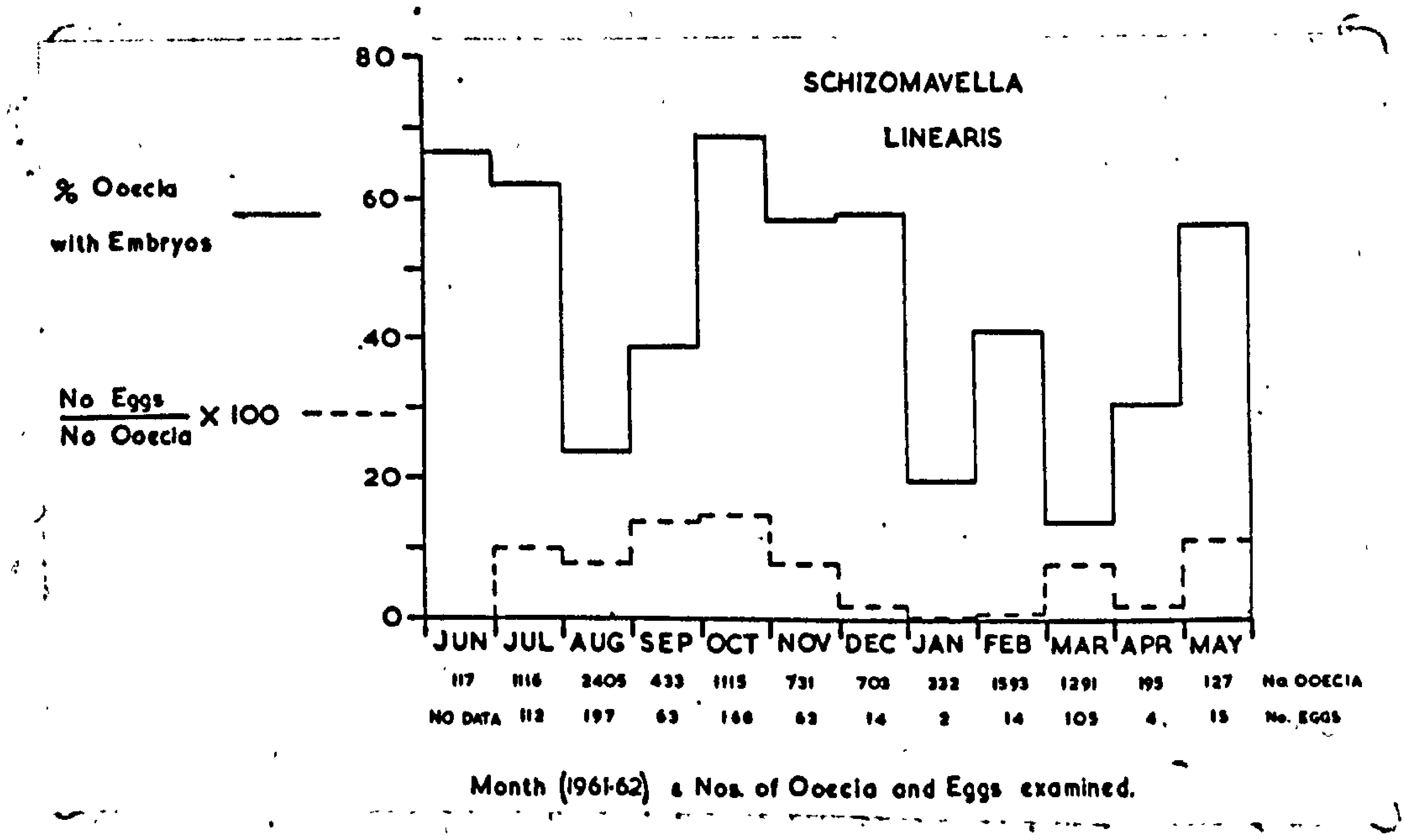


Fig. II.52. <u>Schizomavella</u> <u>linearis</u>. Percentage ooecia with embryos and (No.of eggs: No of ooecia)100 for each month's samples from June 1961 to May 1962.

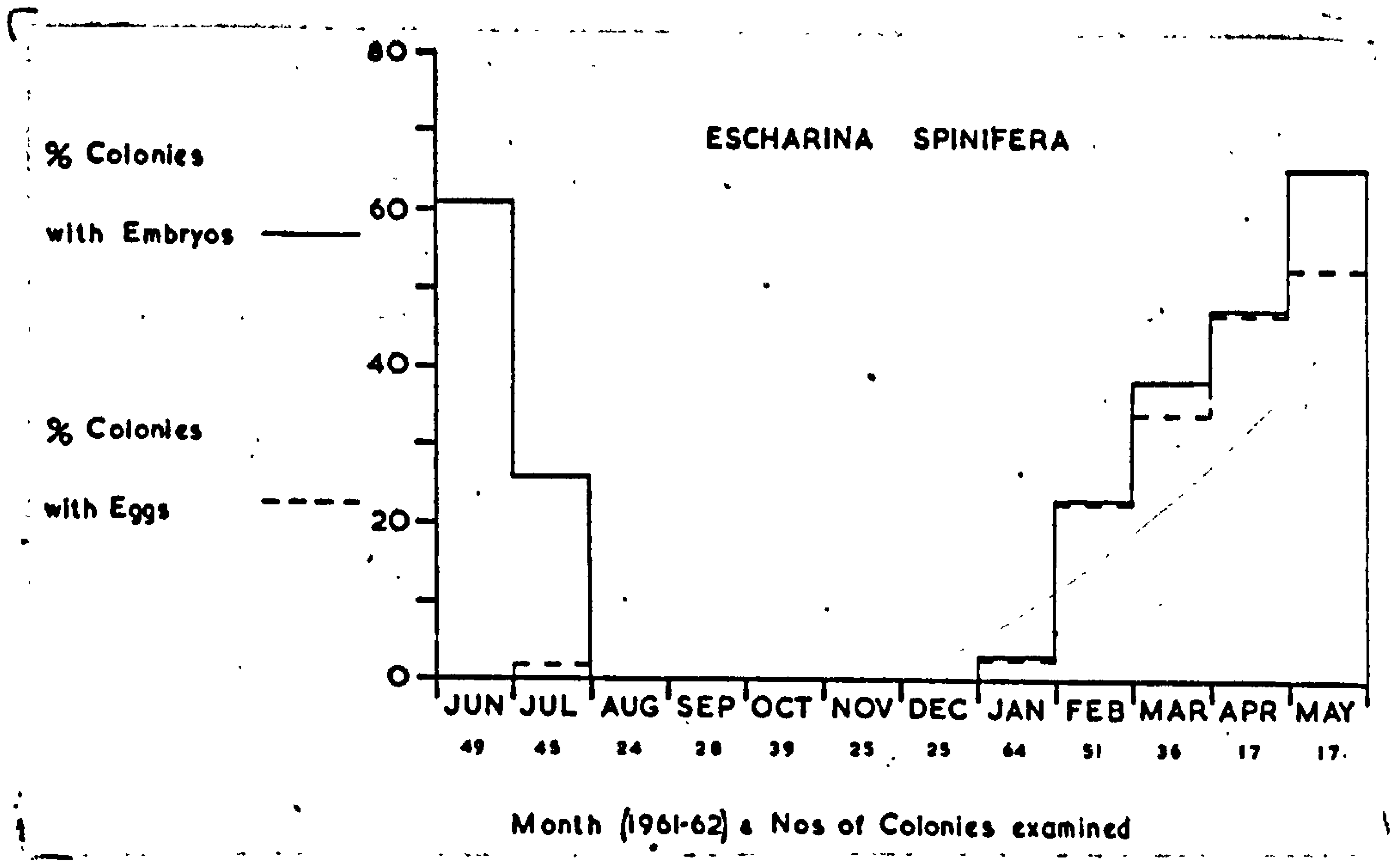


Fig.II.53. Escharina spinifera. Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May 1962.

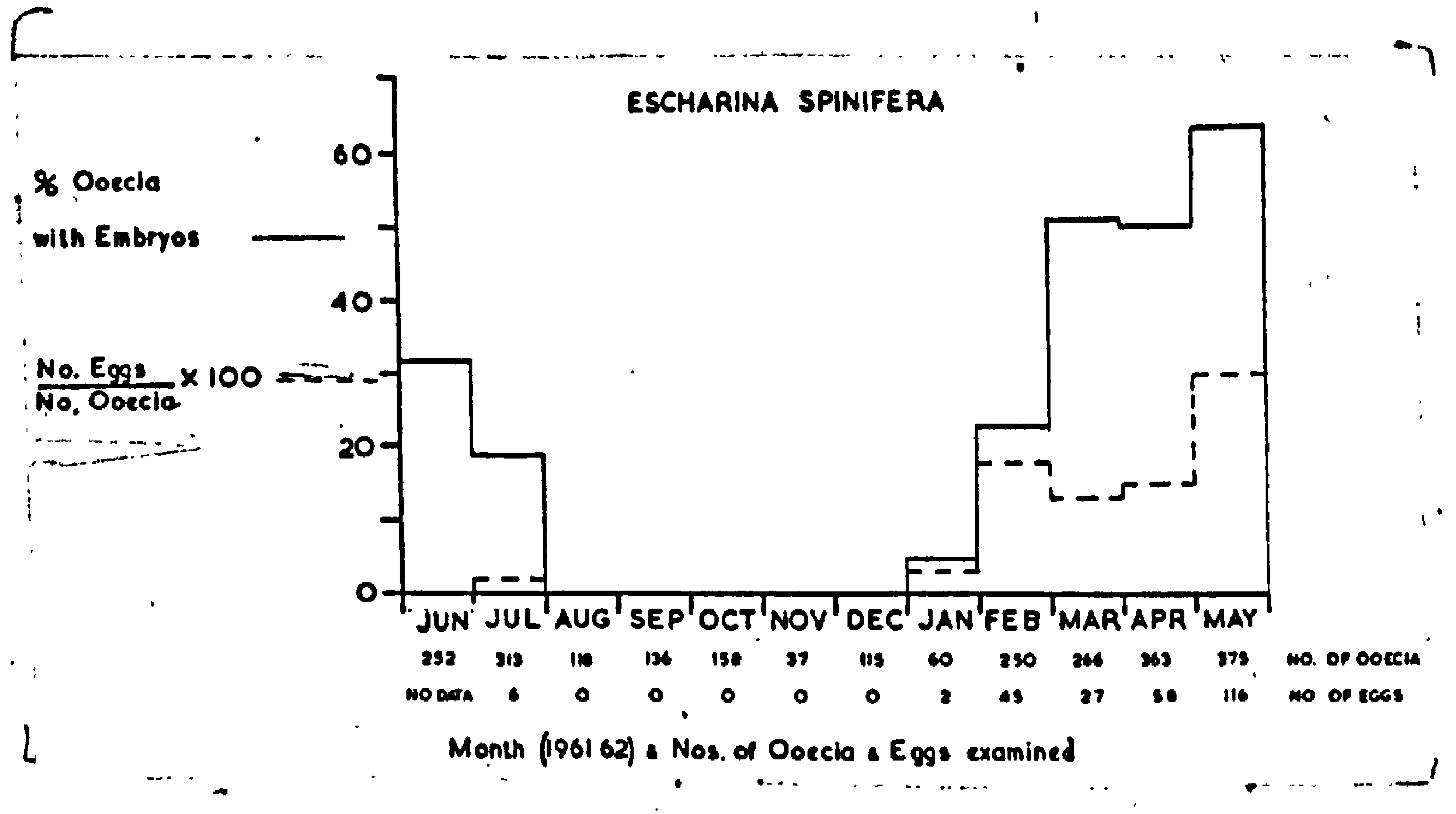


Fig.II. 54. Escharina spinifera. Percentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 foreach month's samples from June 1961 to May 1962.

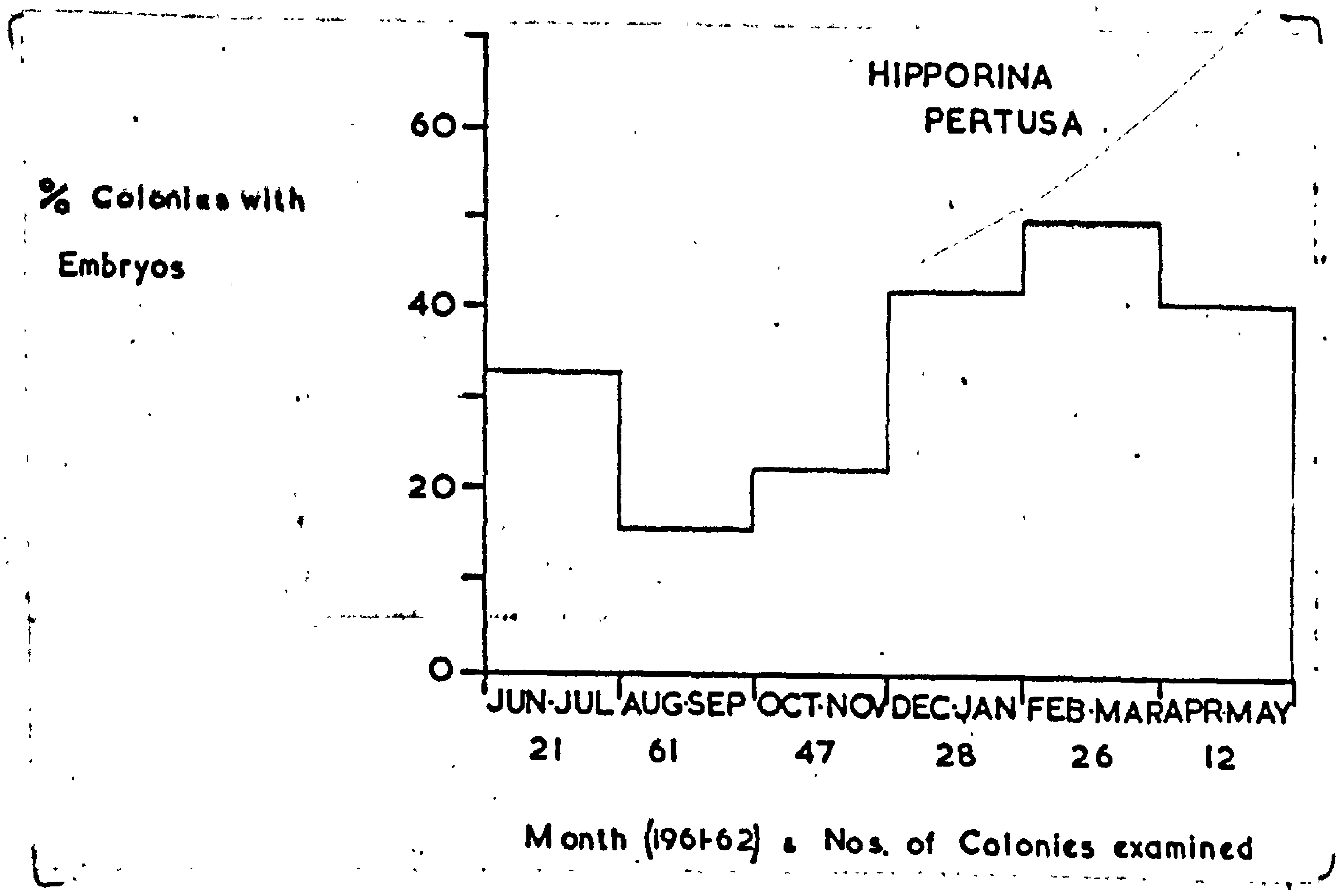


Fig.II. 55. Hipporina pertusa. Percentage colonies with embryos in each two month's samples frpm June 1961 to May 1962.

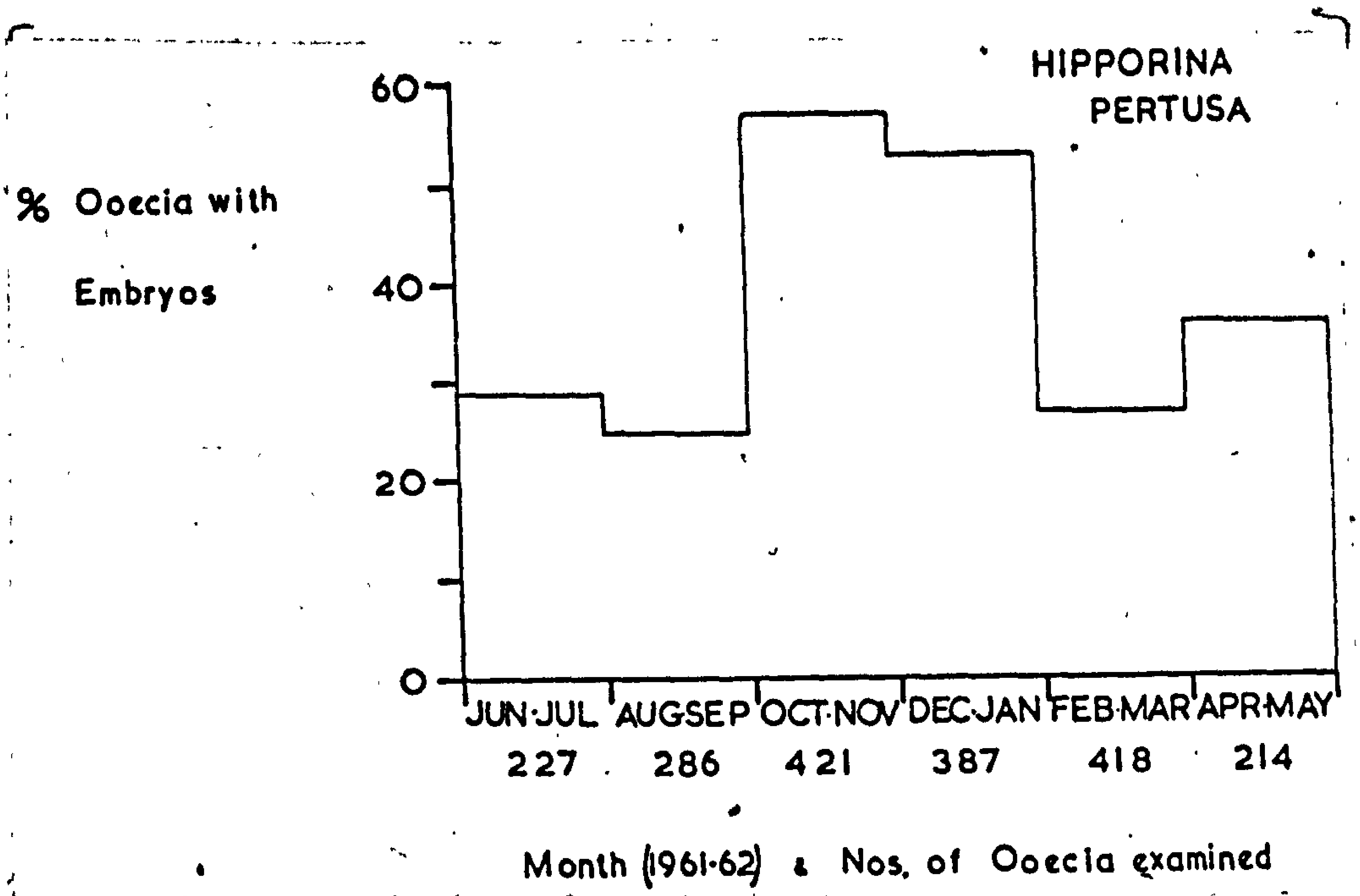


Fig. II.56. Hipporina pertusa. Percentage ooecia containing embryos in each two month's samples from June 1962 to May 1962.

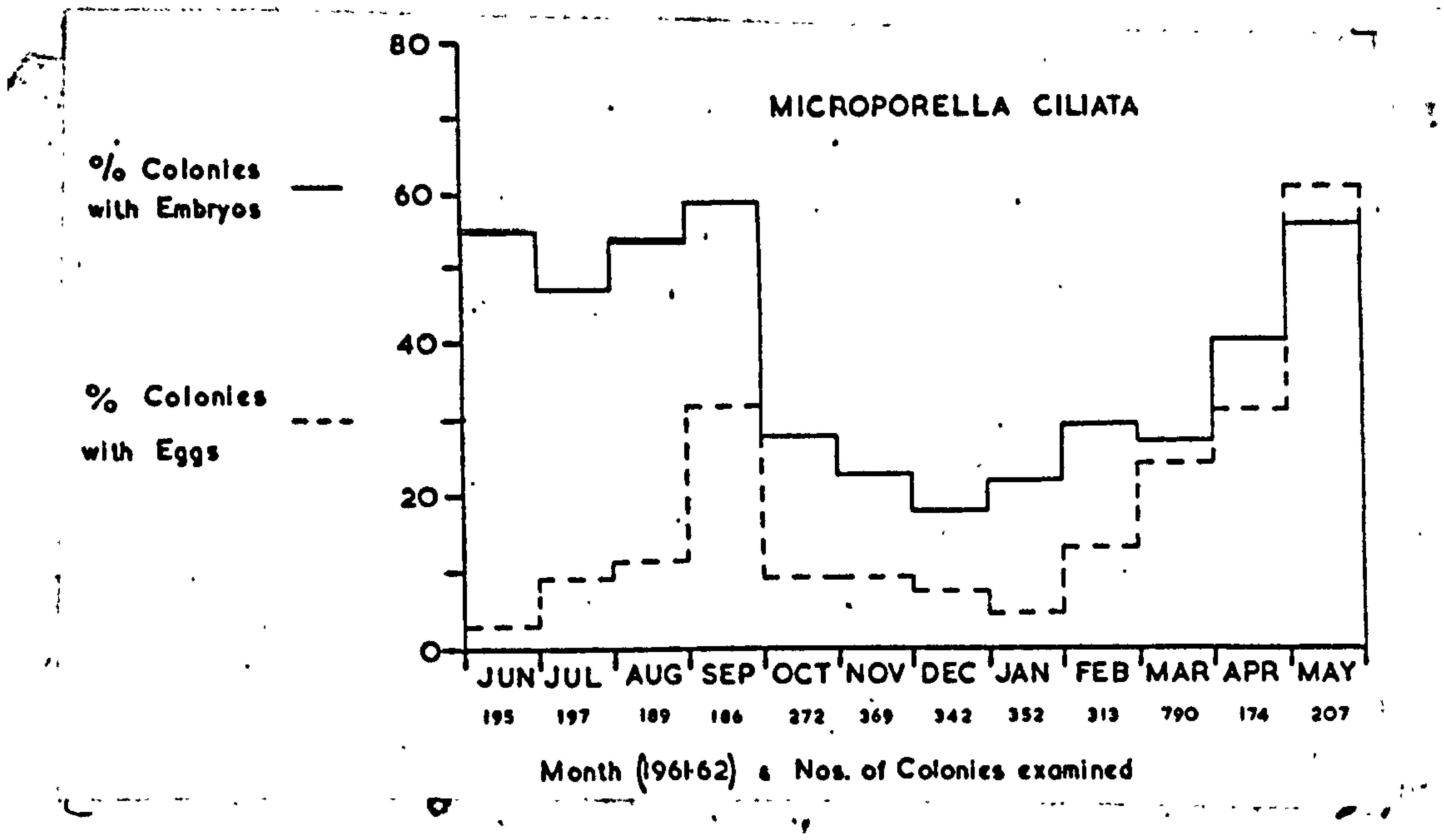


Fig. II.57. <u>Microporella</u> <u>ciliata</u>. Percentage colonies with eggs and percentage with embryos in each month's samples from June 1961 to May 1962

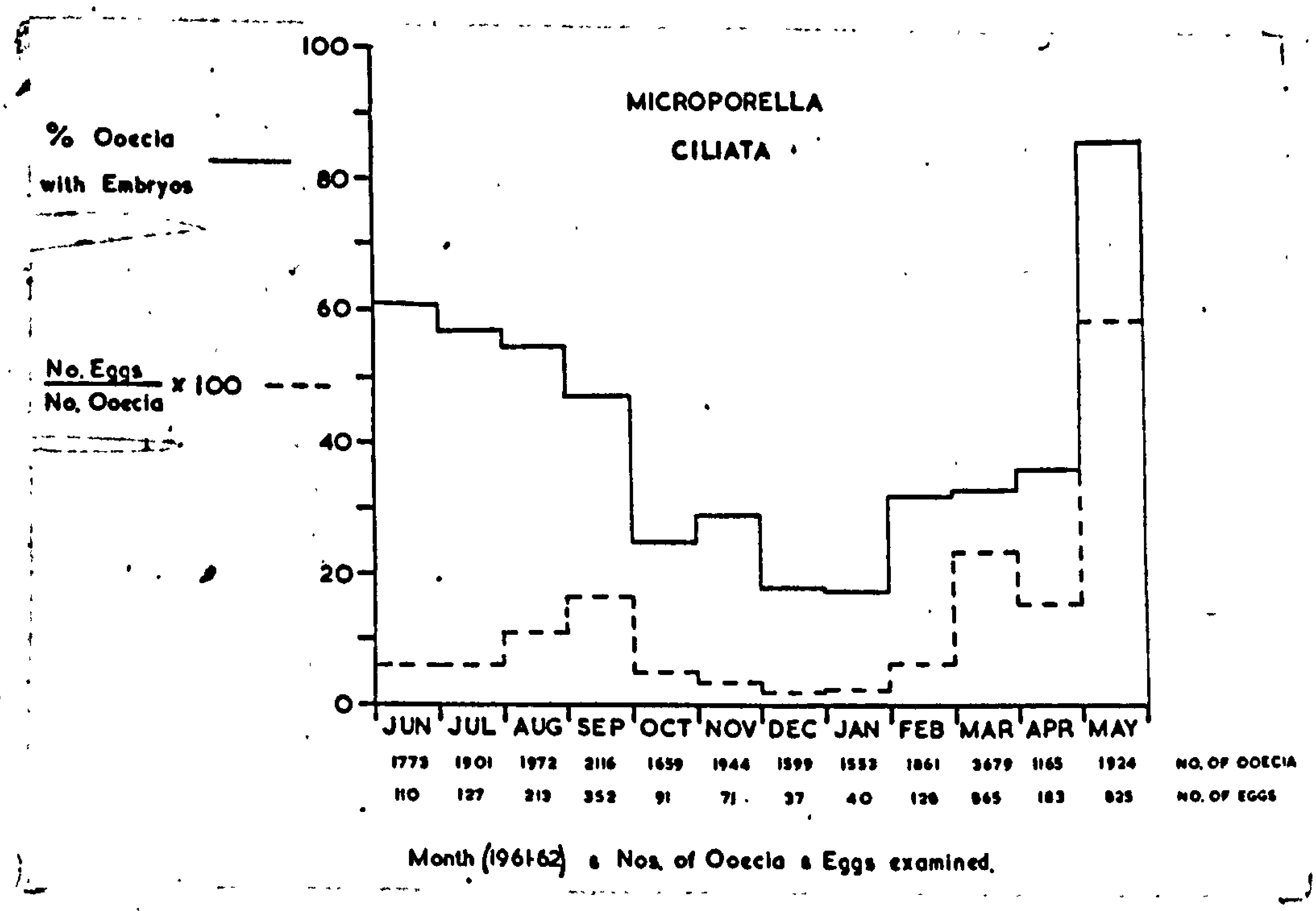


Fig. II.58. <u>Microporella</u> <u>ciliata</u>. Percentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 for each month's samples from June 1961 to May 1962.

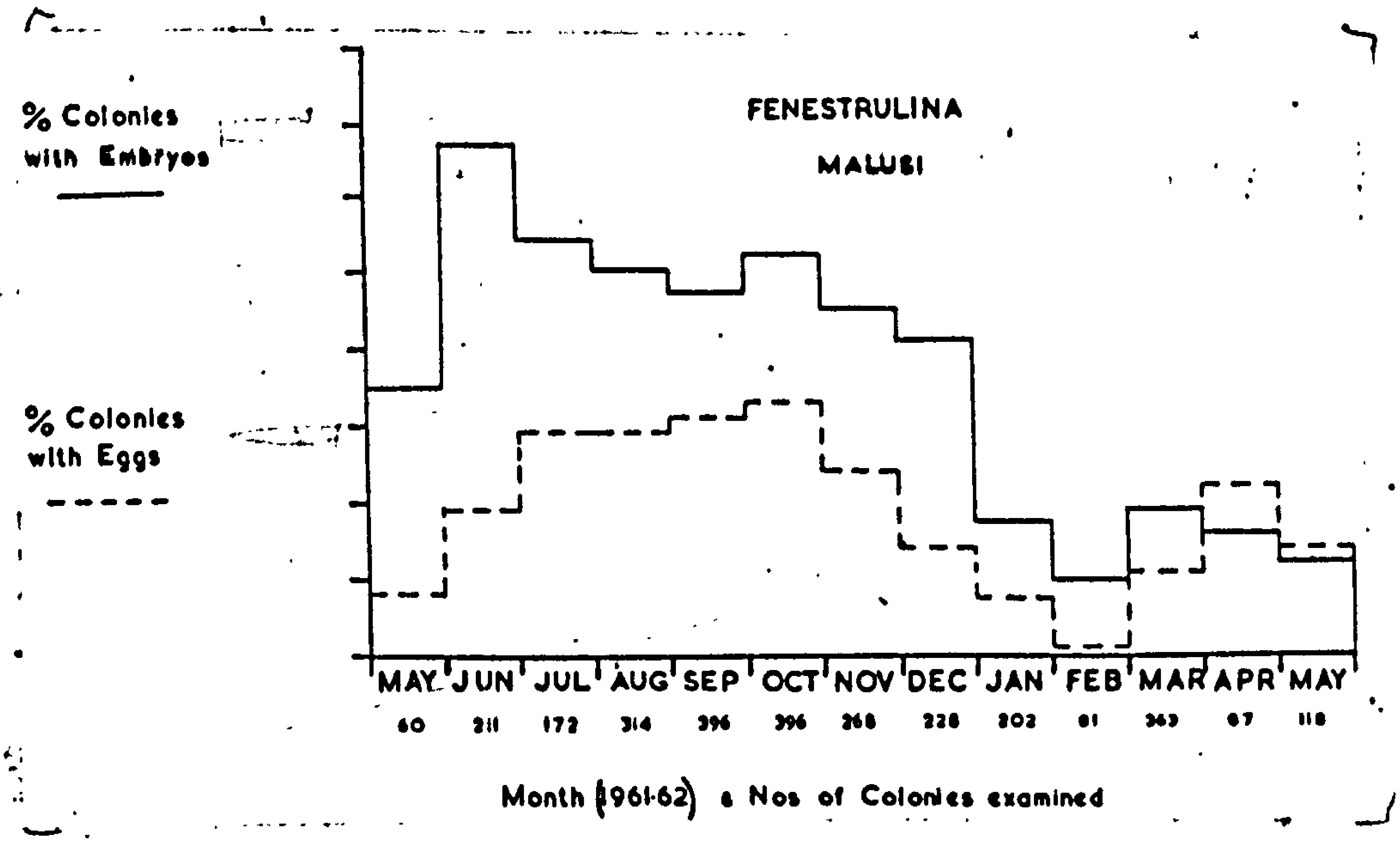


Fig. II. 59. Fenestrulina malusi. Percentage colonies with embryos and percentage with eggs in each month's samples from May 1961 to May 1962

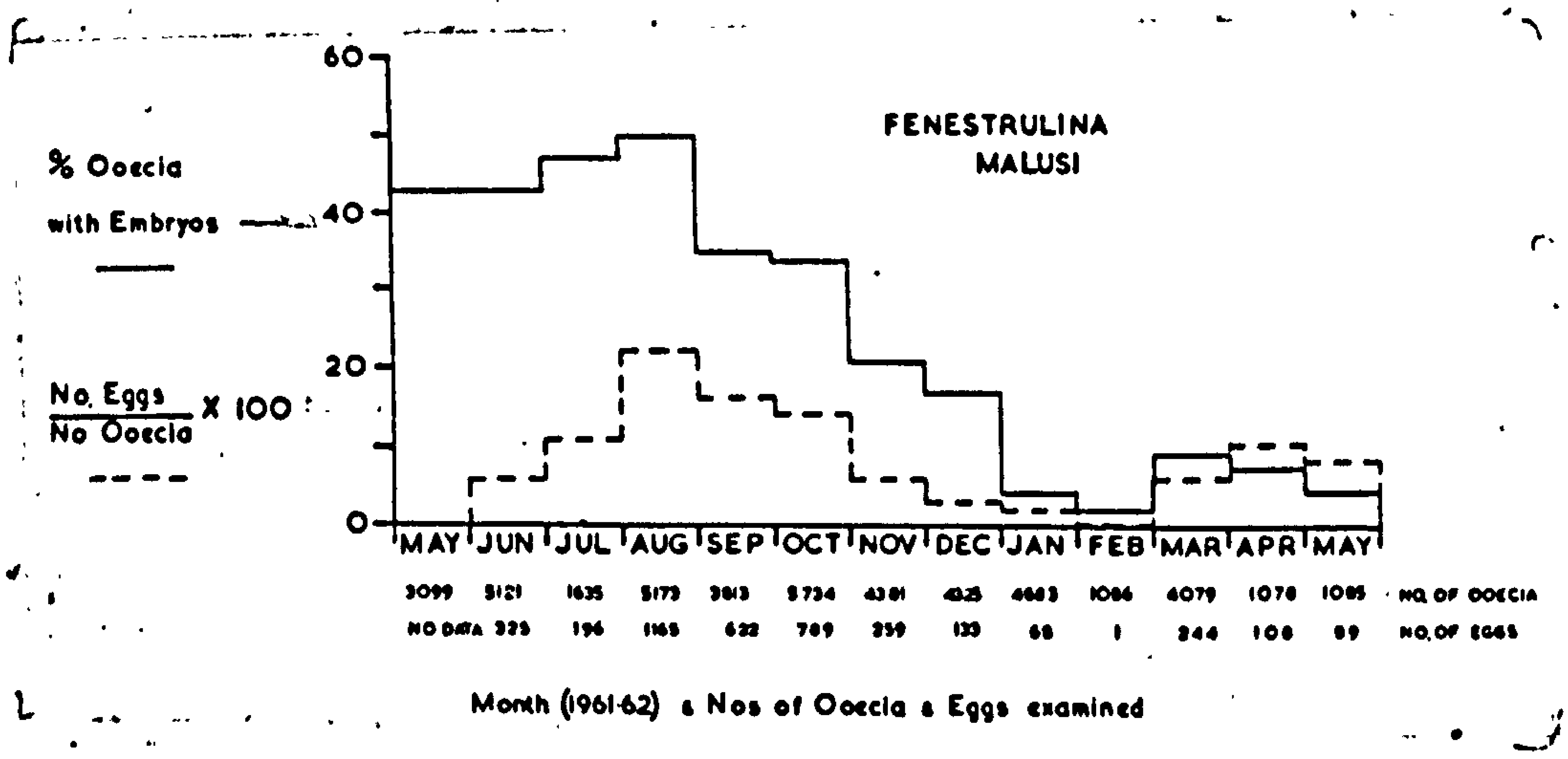


Fig.II.60. Fenestrulina malusi. Percentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 for each month's sample from May 1961 to May 1962.

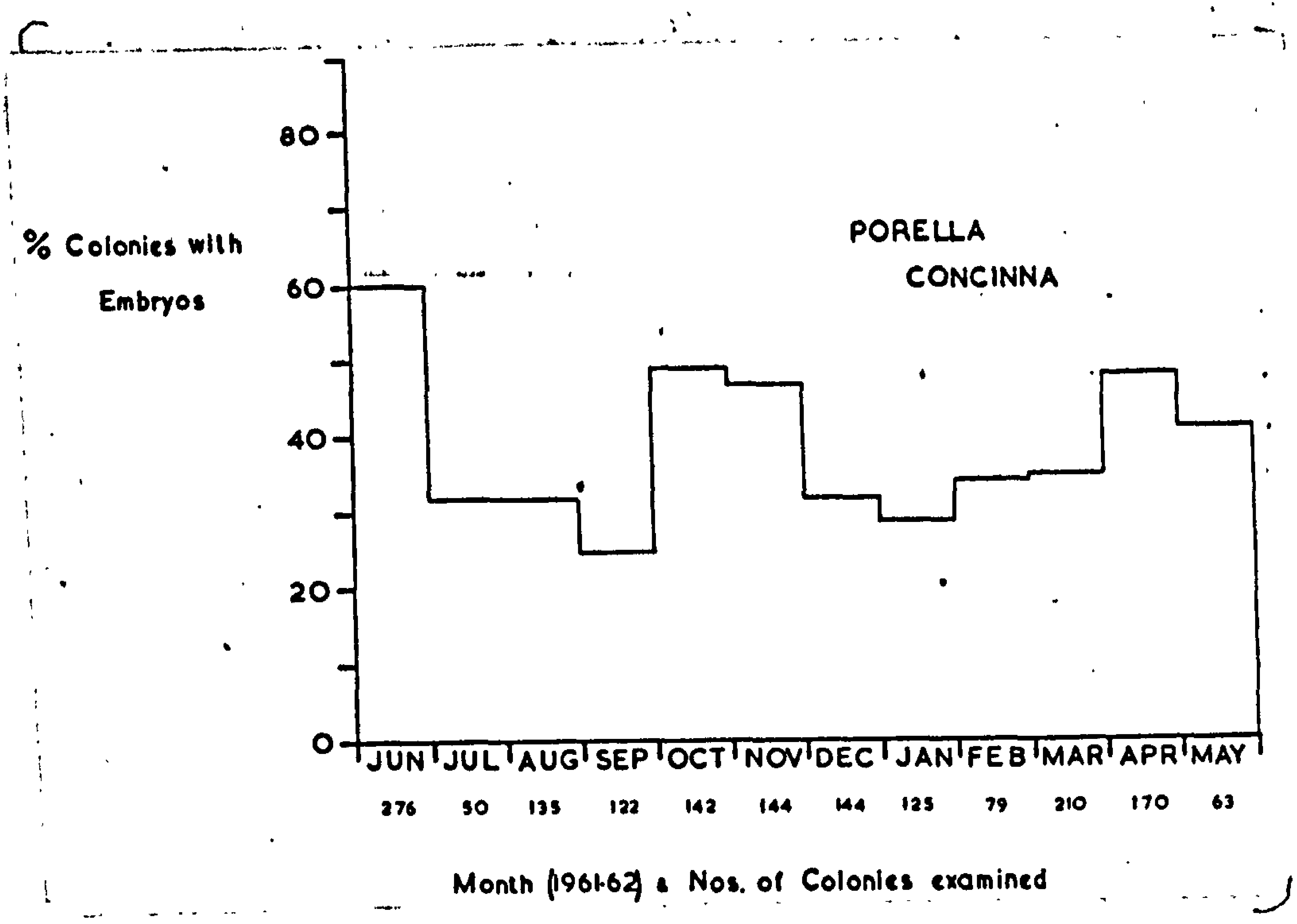


Fig. II.61 Porella concinna. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

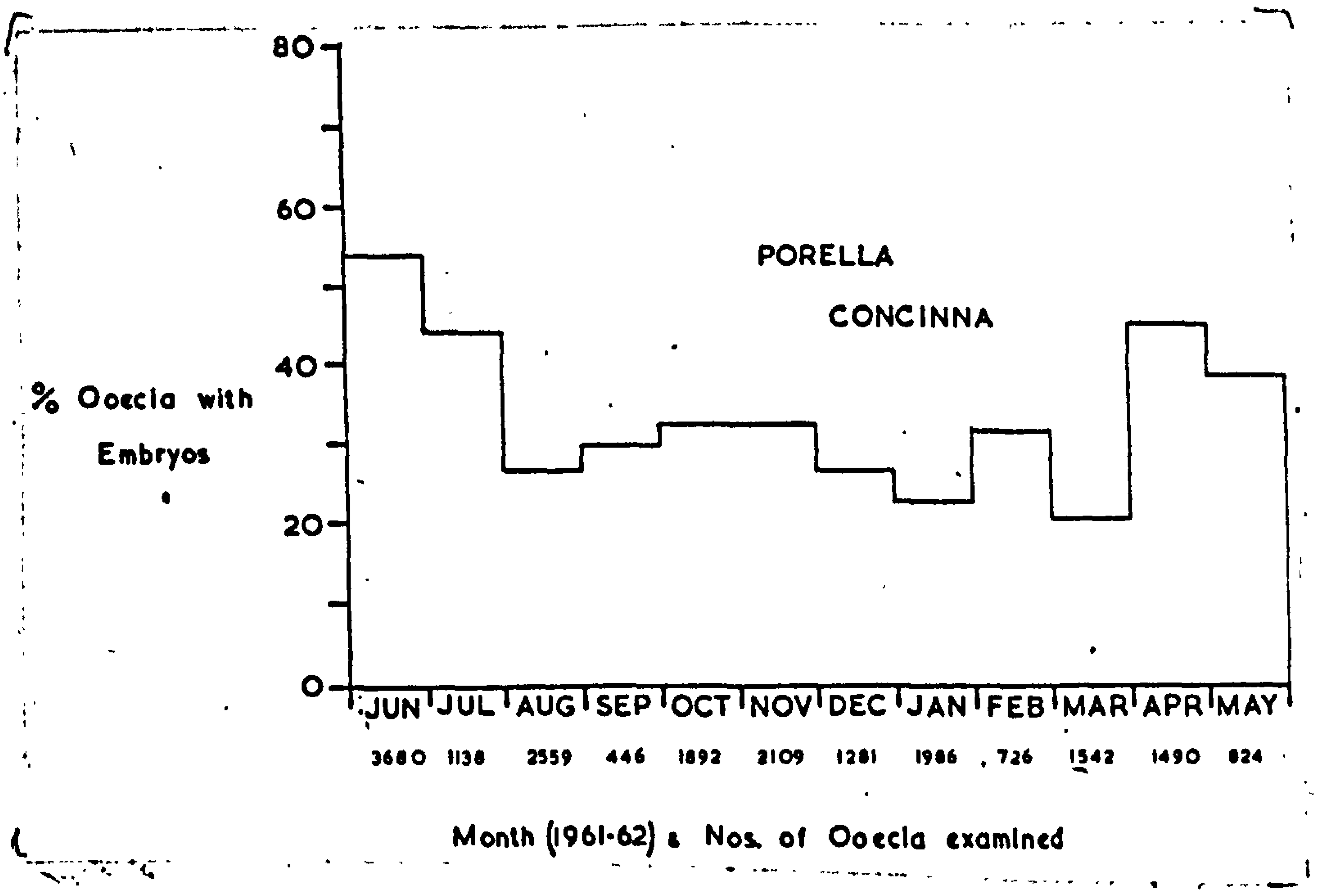


Fig.II.62. Porella concinna. Percentage oöecia containing embryos in each month's samples from June 1961 to May 1962.

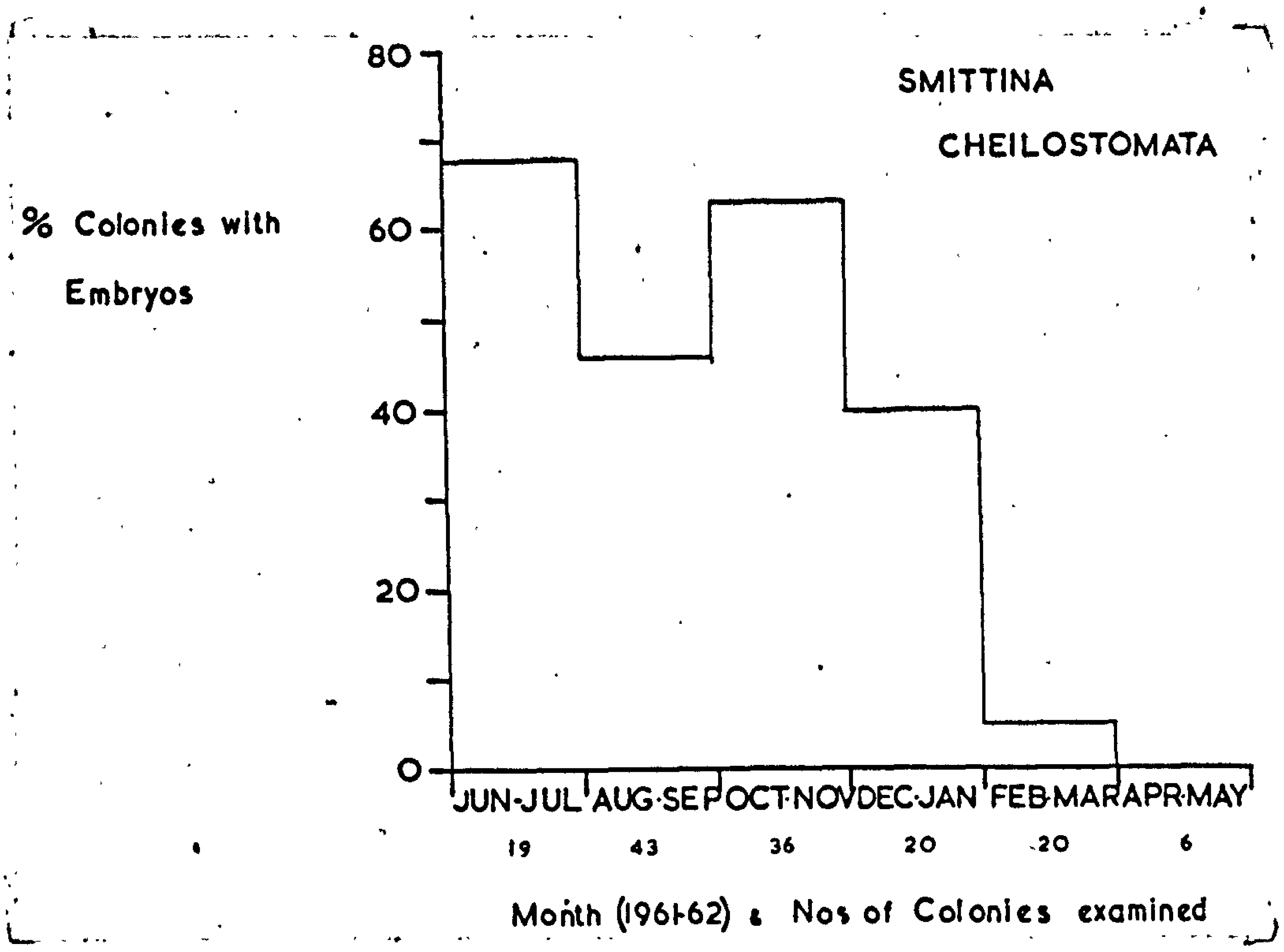


Fig.II. 63. Smittina cheilostomata. Percentage colonies with embryos in each two month's samples from June 1961 to May 1962.

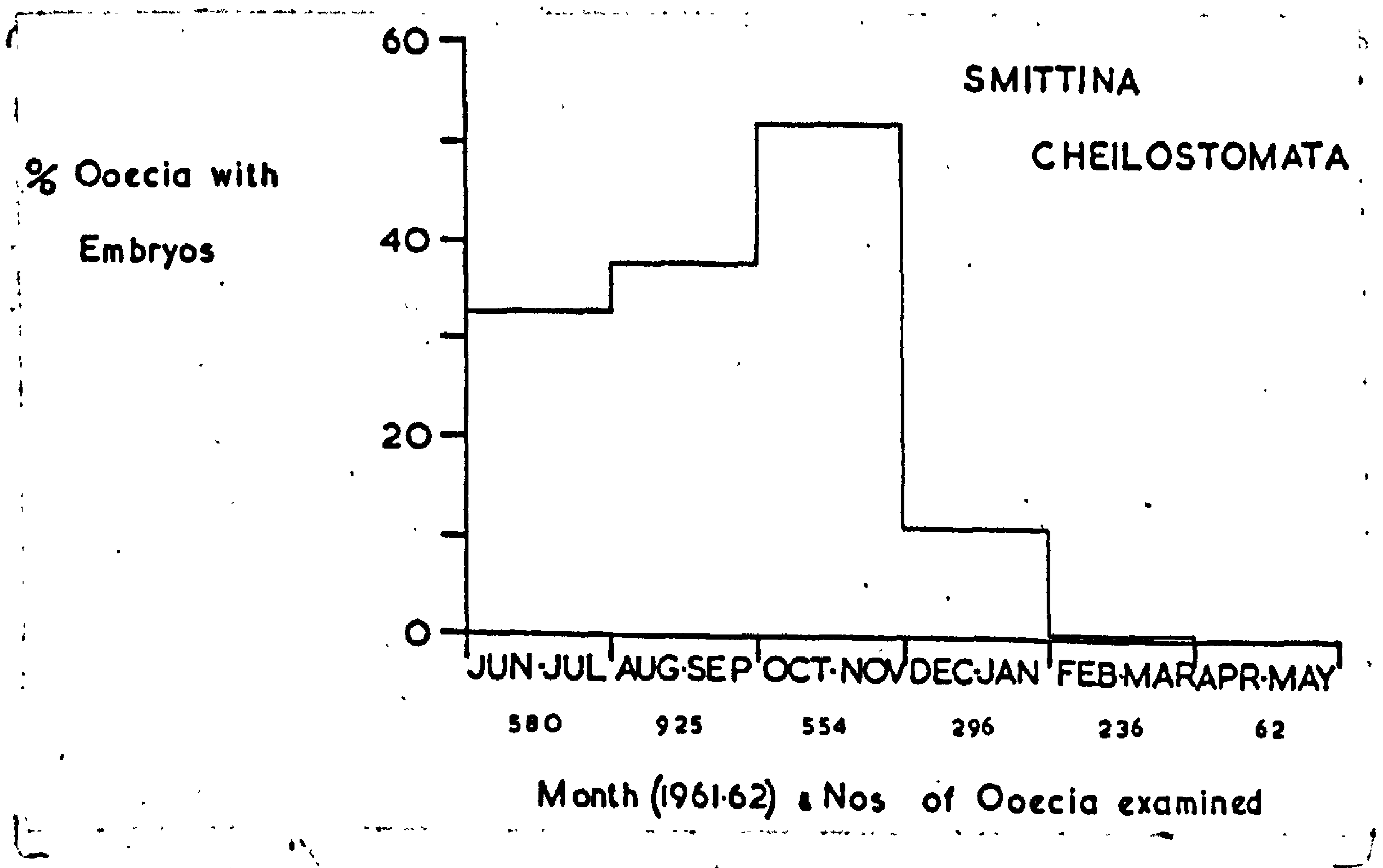


Fig. II.64. Smittina cheilostomata. Percentage ooecia containing embryos in each two month's samples from June 1961 to May 1962.

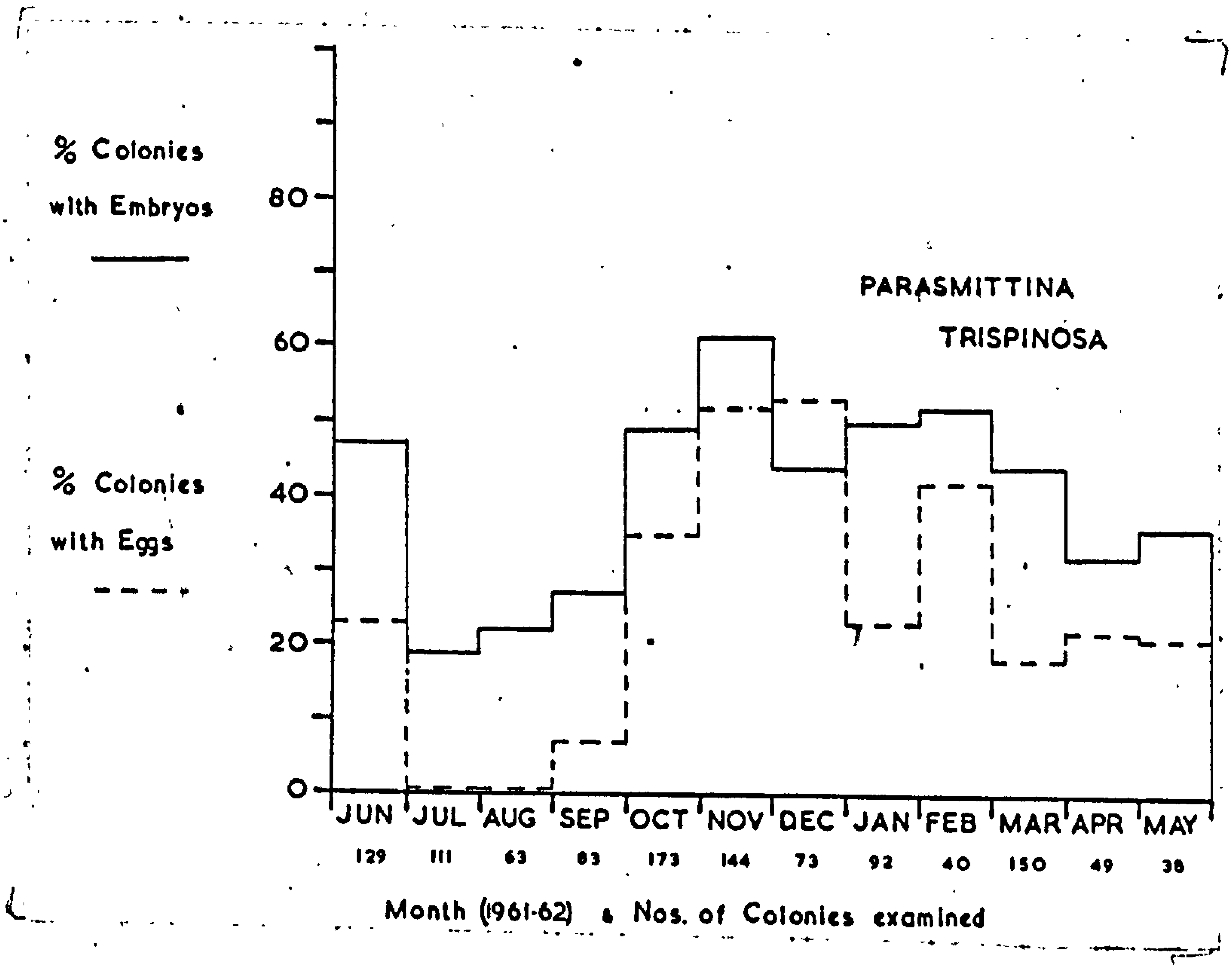


Fig II. 65. Parasmittina trispinosa. Percentage colonies with embryos and percentage with eggs in each month's samples from June 1961 to May 1962.

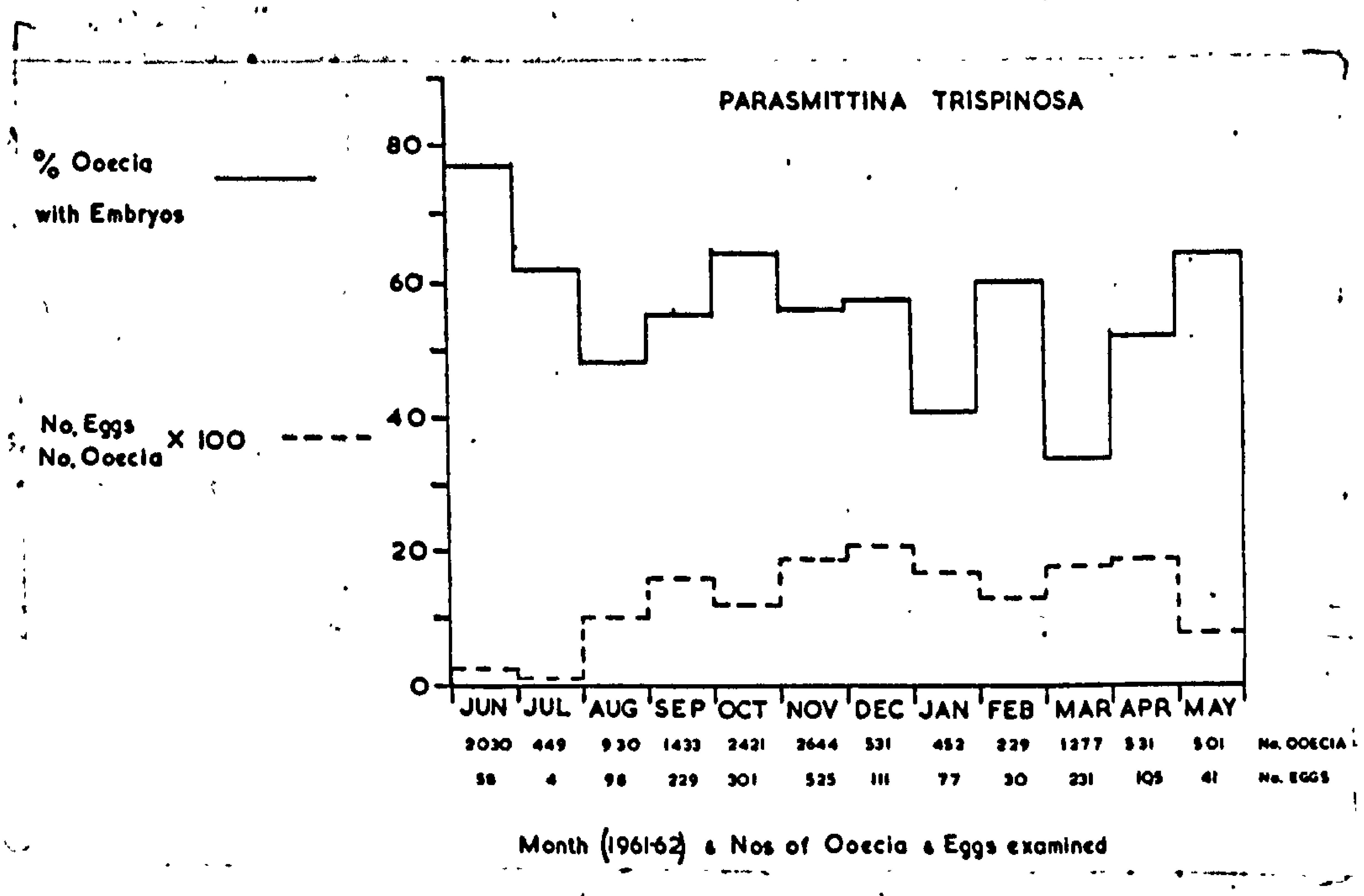


Fig.II.66. Parasmittina trispinosa. Percentage ooecia containing embryos and (No. of eggs: No. of ooecia)100 for each month's samples from June 1961 to May 1962.

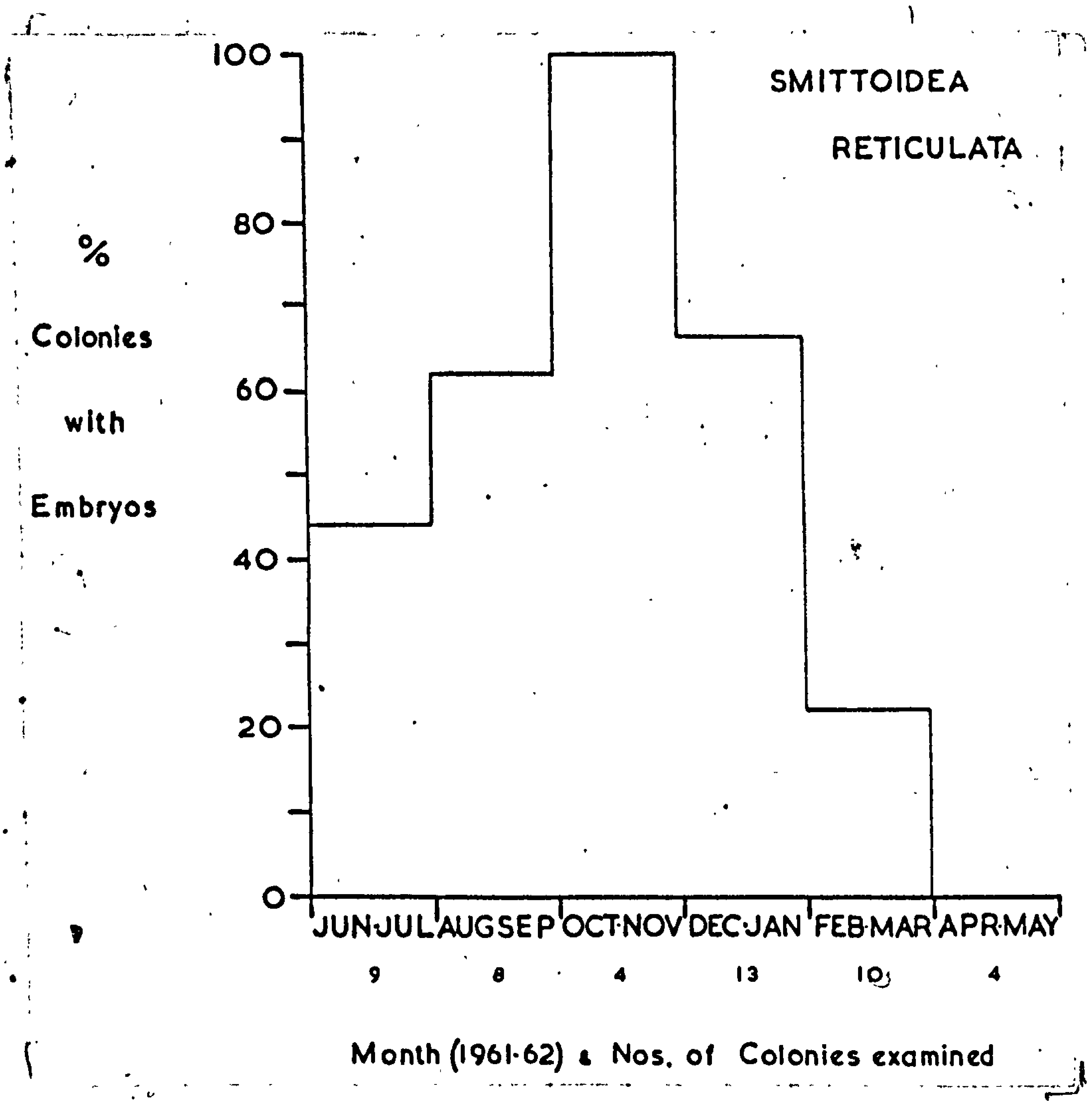


Fig.II.67. Smittoidea reticulata. Percentage colonies with embryos in each two month's samples from June 1961 to May 1962.

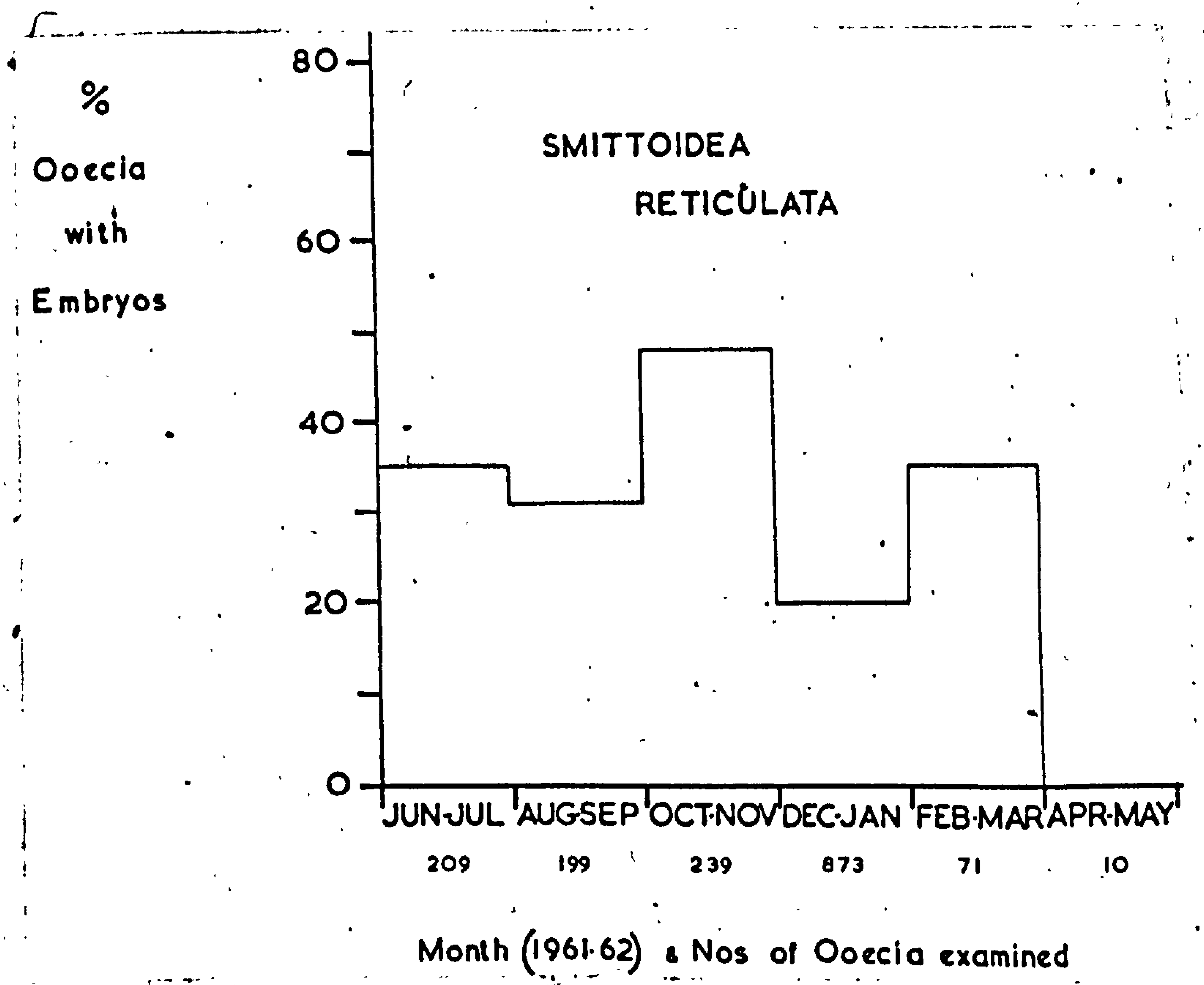


Fig.II.68. Smittoidea reticulata. Percentage ooecia containing embryos in each two month's samples from June 1961 to May 1962.

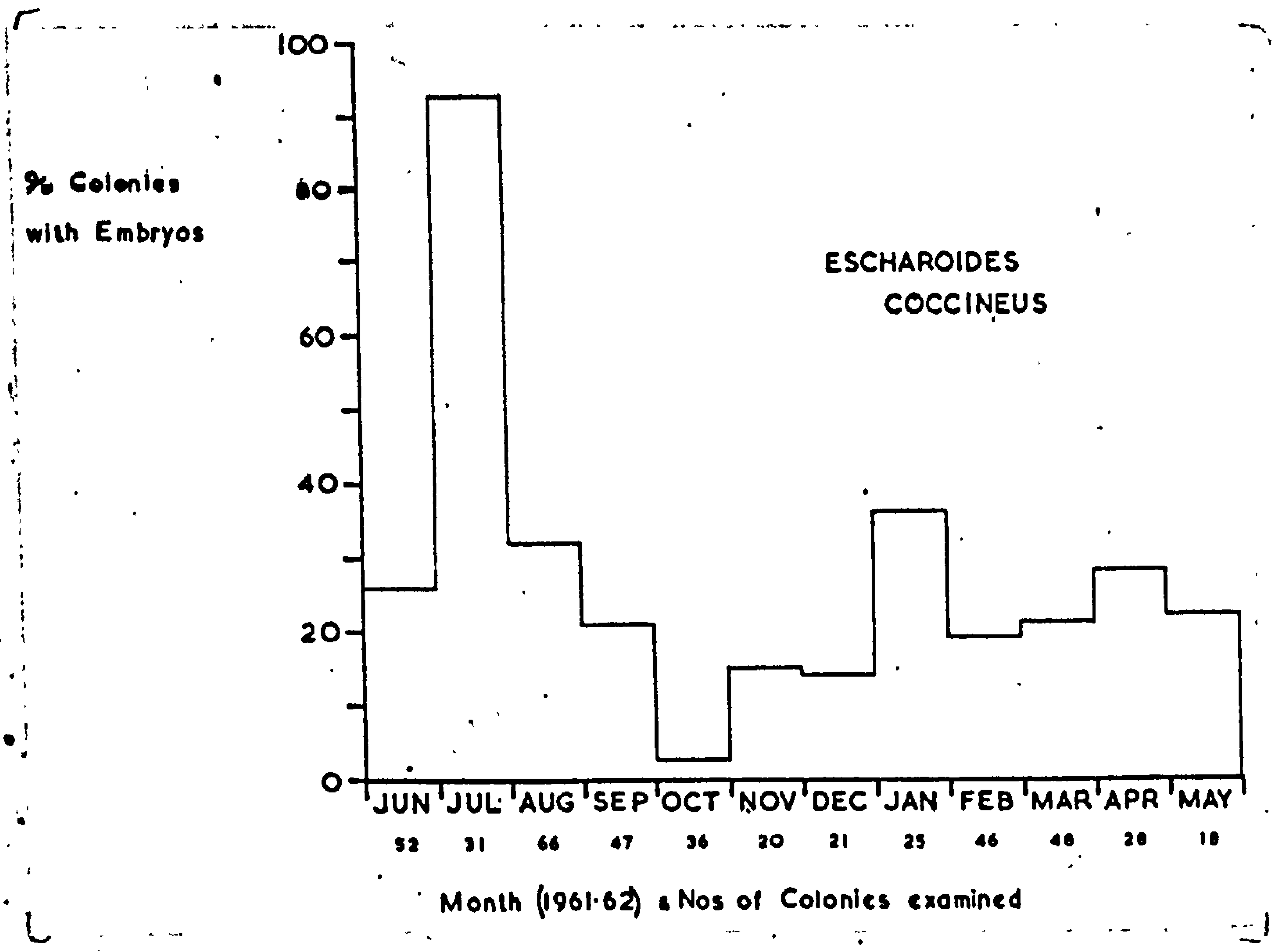


Fig. II. 69. Escharoides coccineus. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

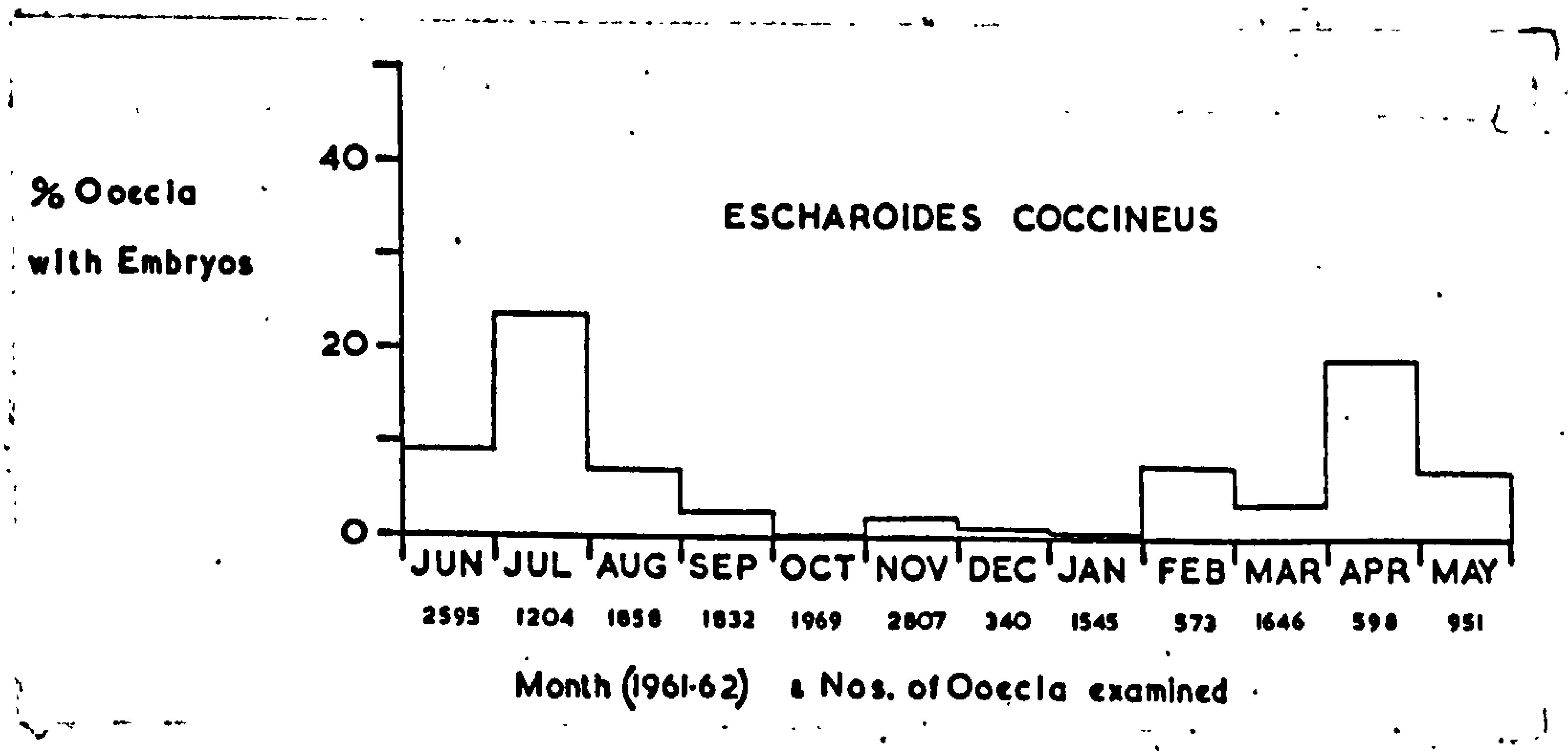


Fig. II. 70. Escharoides coccineus. Percentage ooecia containing embryos in each month's samples from June 1961 to May 1 962.

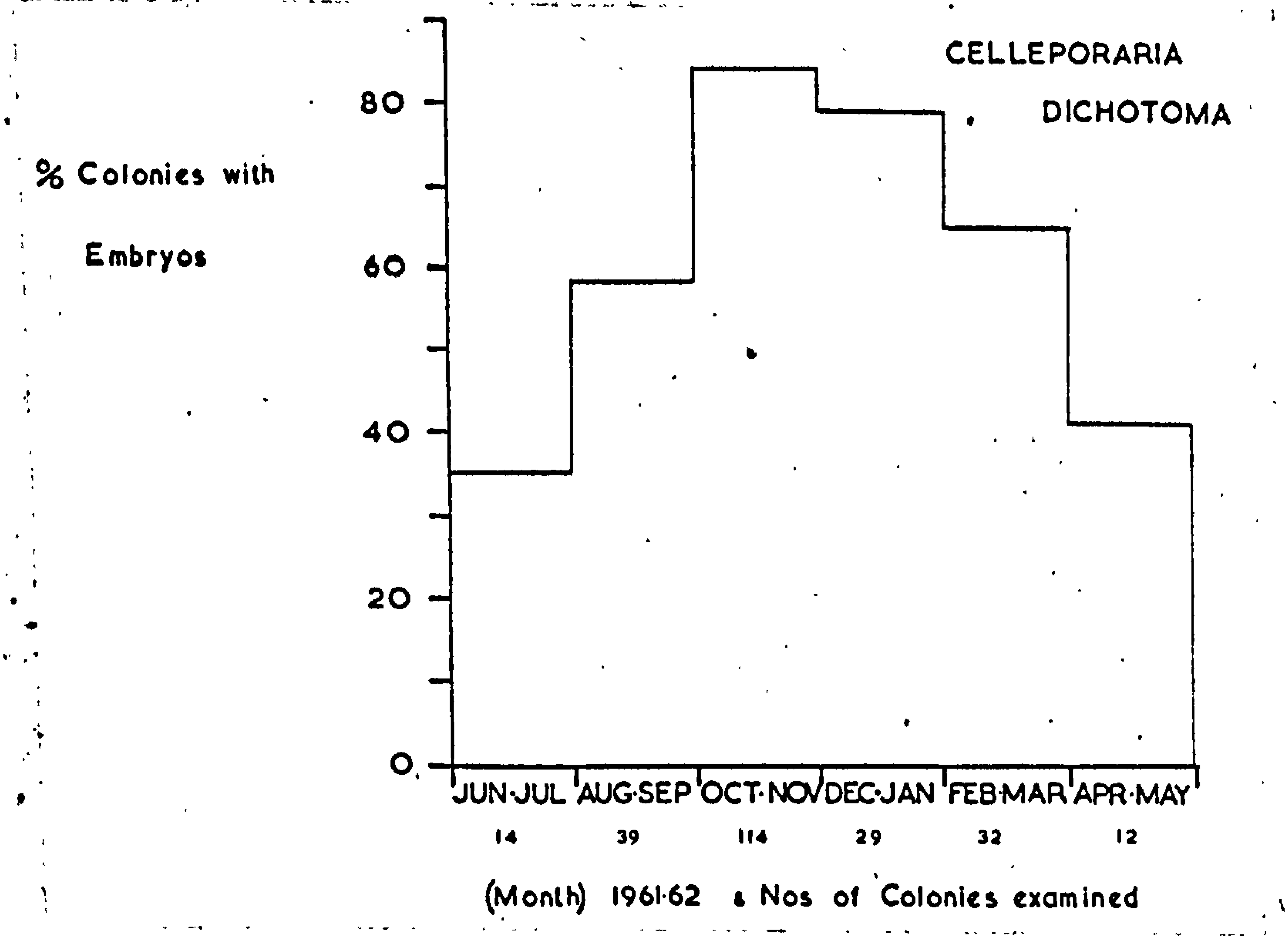


Fig.II. 71. Celleporaria dichotoma. Percentage colonies with embryos in each two months samples from June 1961 to May 1962.

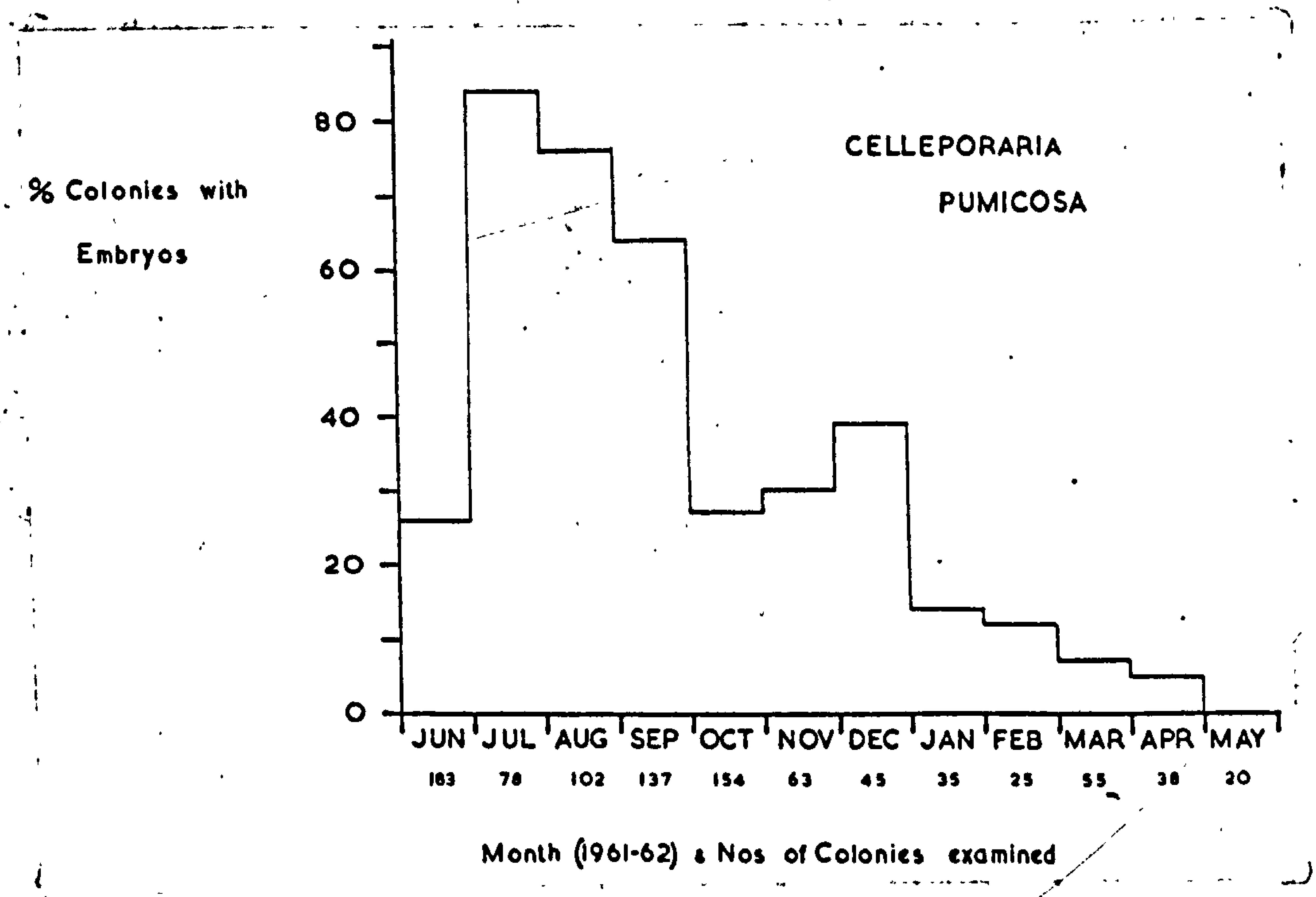


Fig. II.72. Celleporaria pumicosa. Percentage colonies with embryos in each month's samples from June 1961 to May 1962,

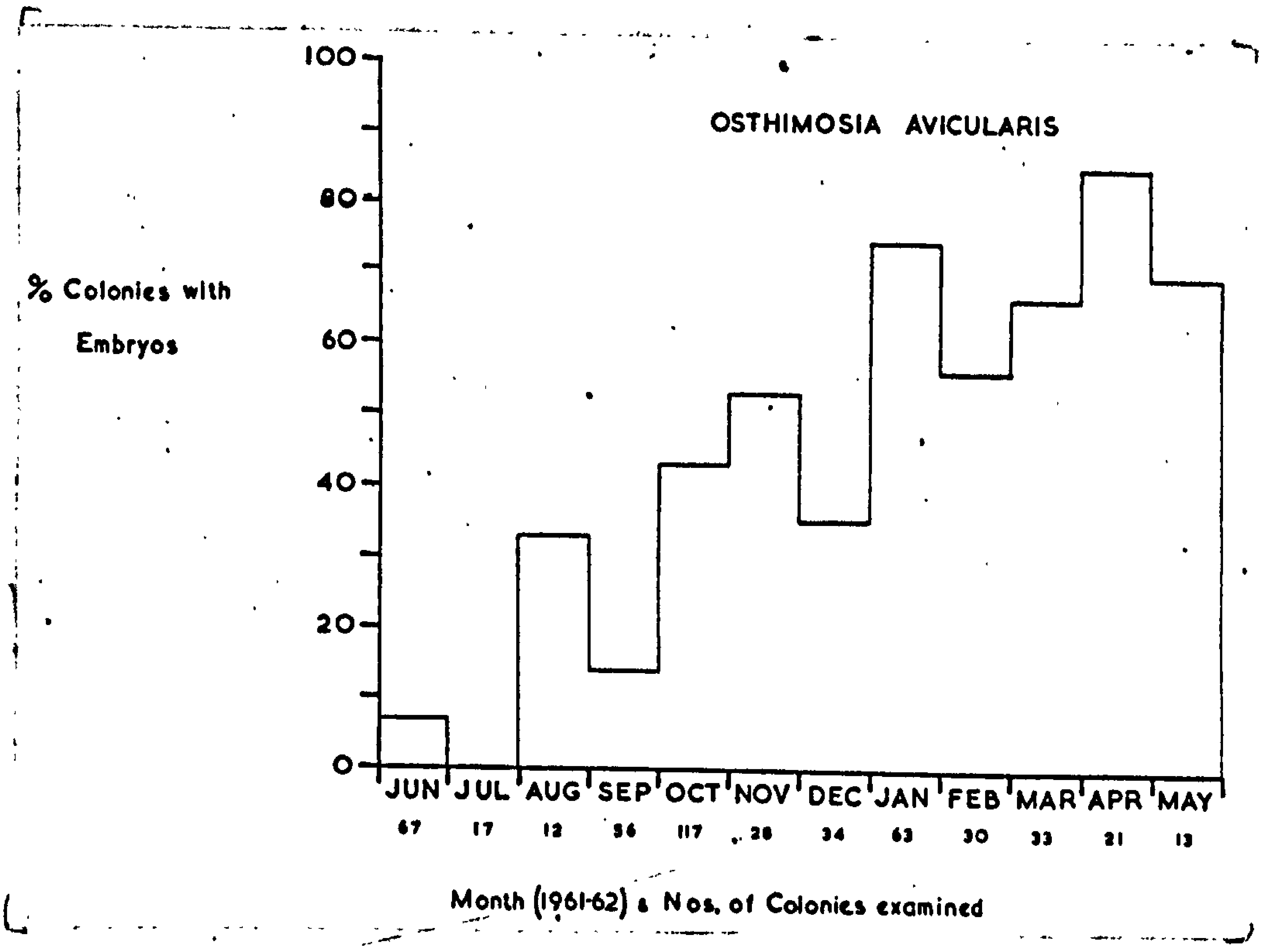


Fig. II. 73. Osthimosia avicularis. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

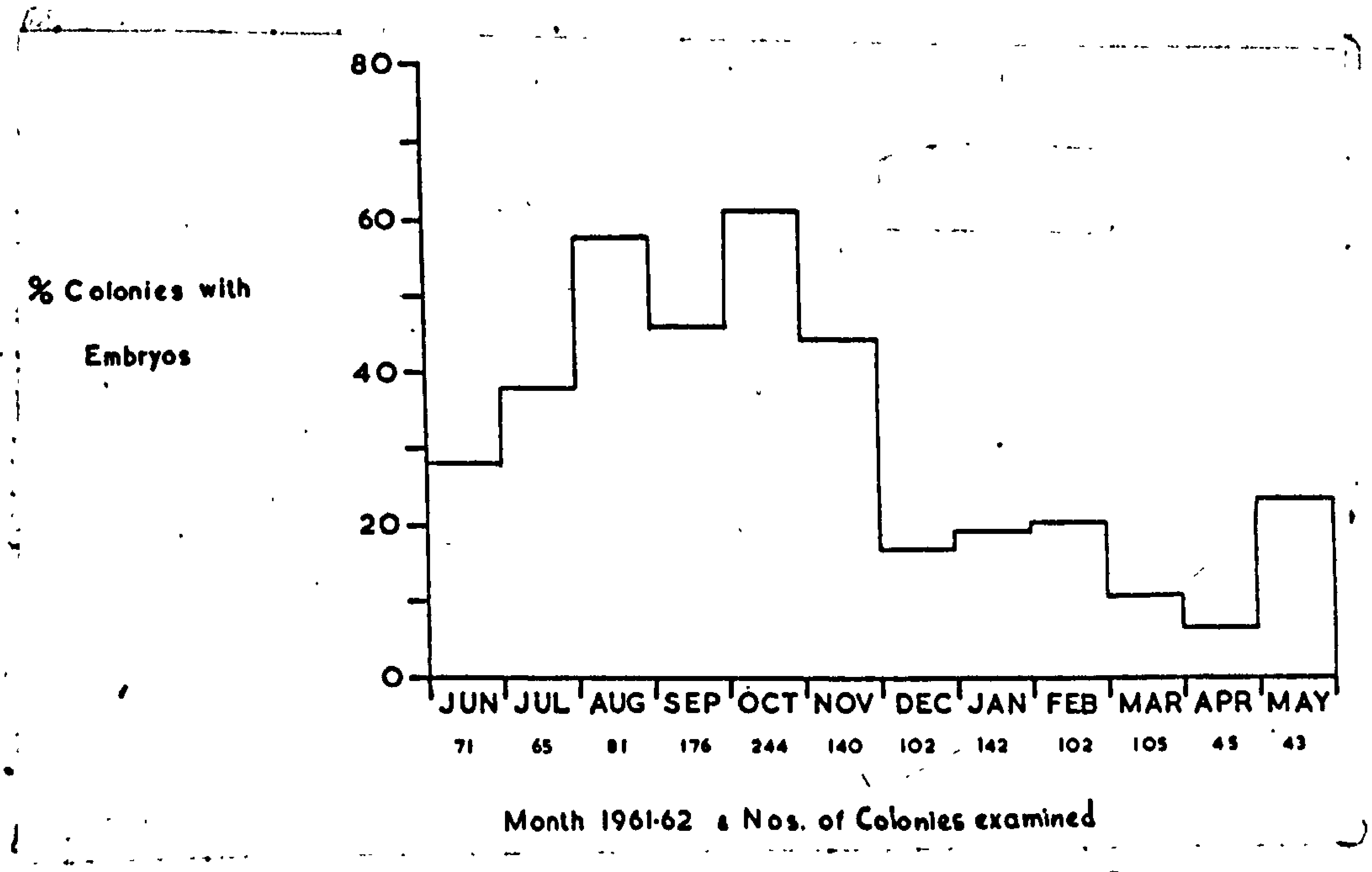


Fig. II. 74. Celleporina costazii. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

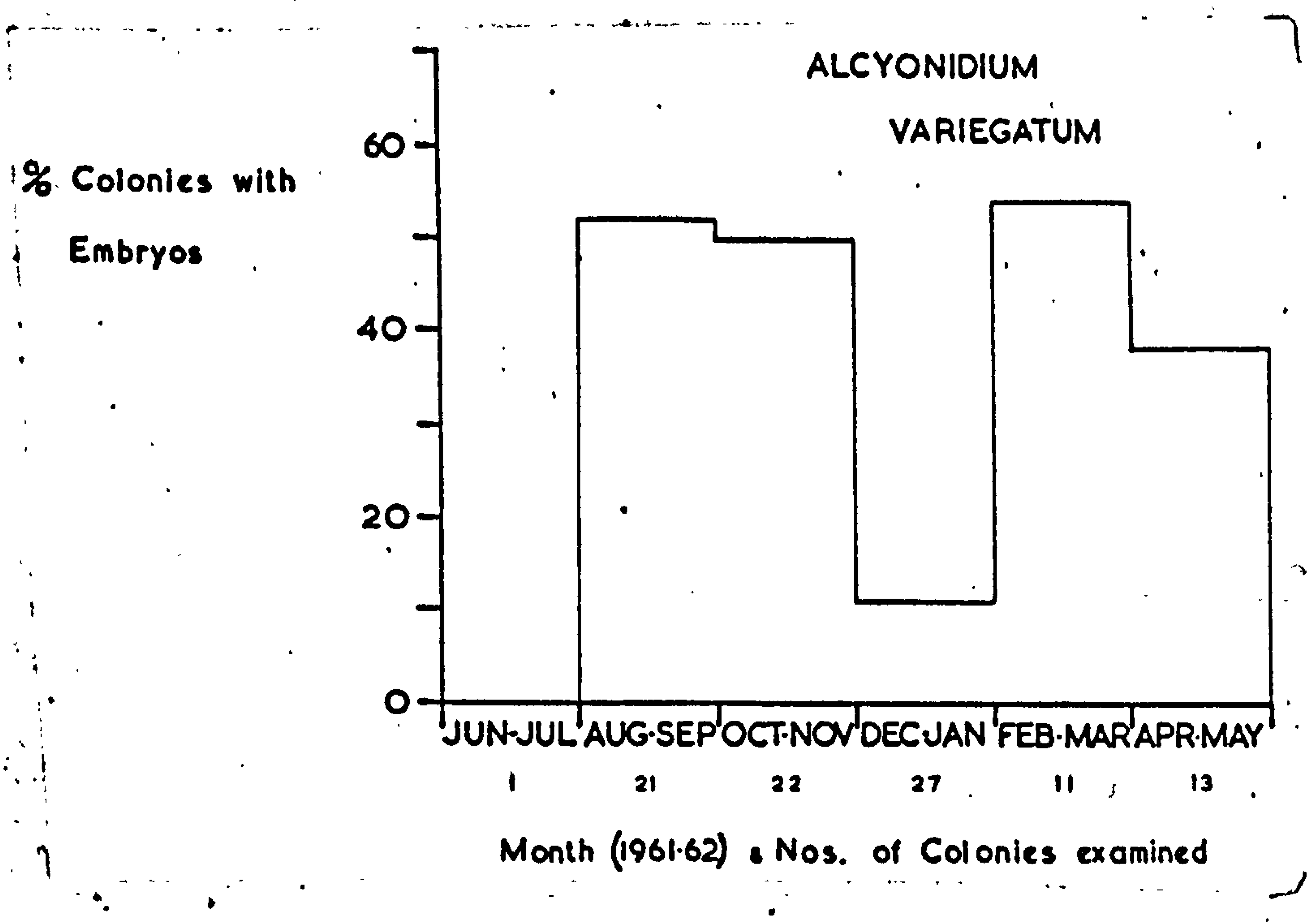


Fig. II.75. Alcyonidium variegatum. Percentage colonies with embryos in each two month's samples from June 1961 to May 1962.

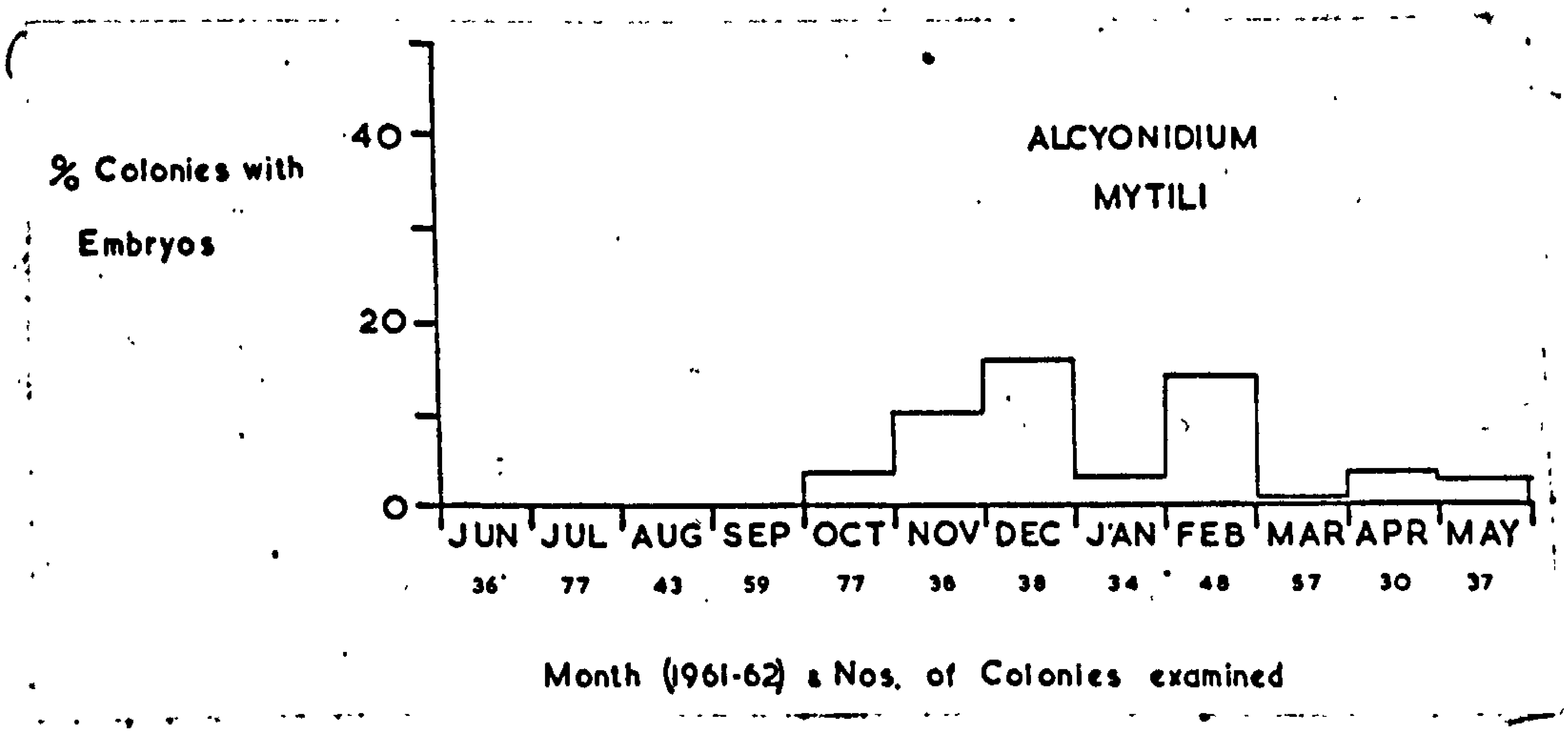


Fig.II. 76. Alcyonidium mytili. Percentage colonies with embryos in each month's samples from June 1961 to May 1962.

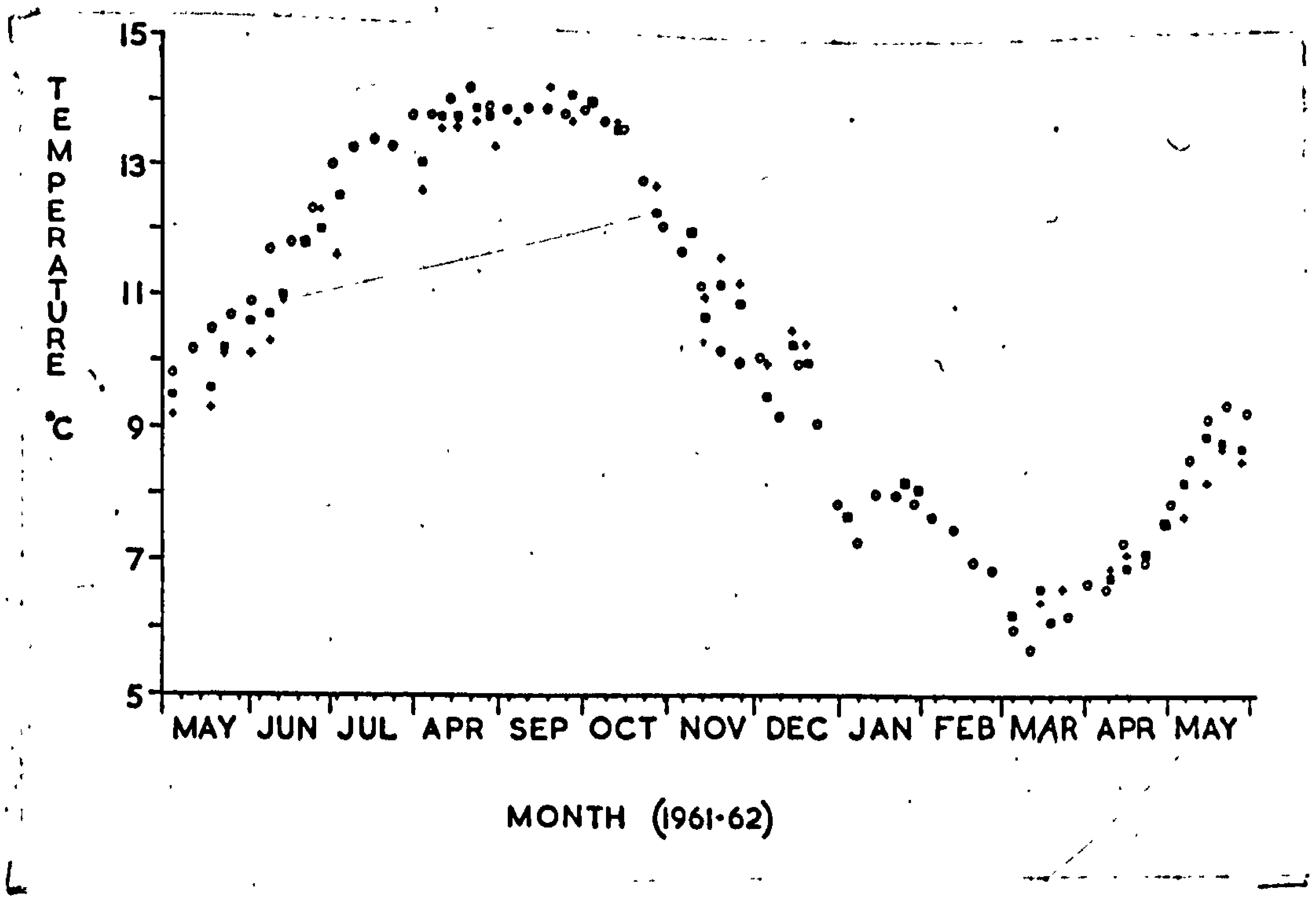
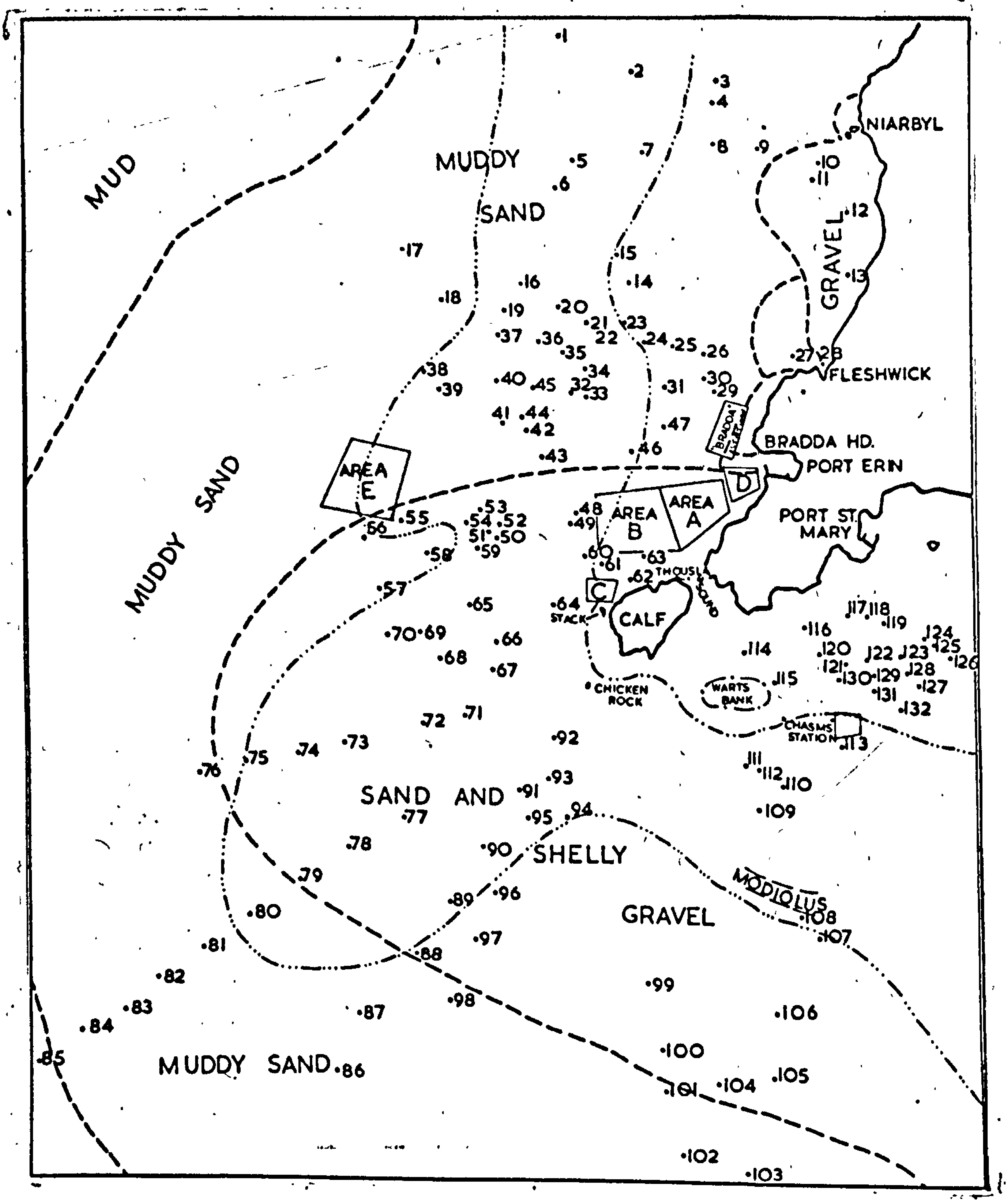
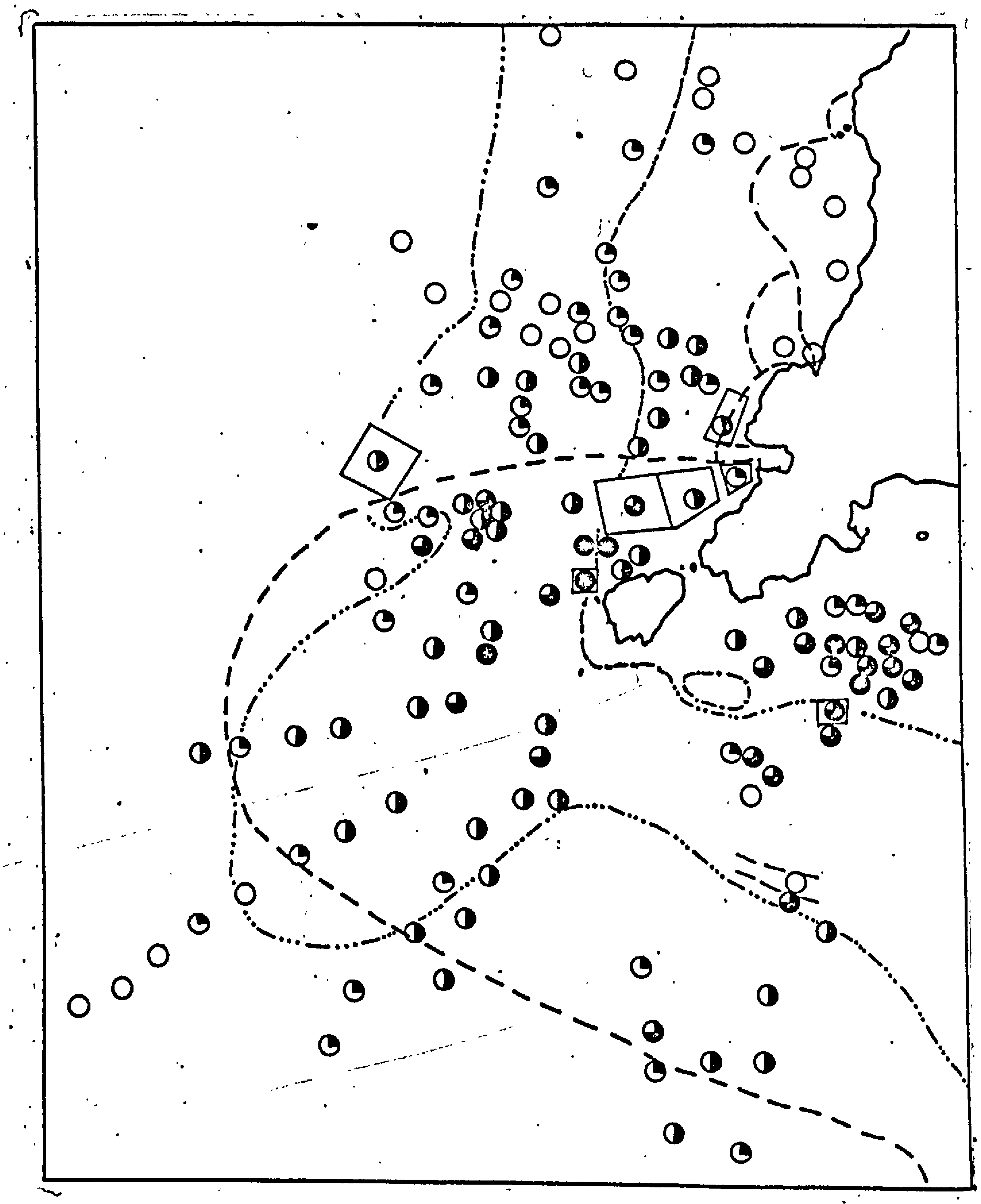


Fig. II. 77. Sea temperatures from May 1961 to May 1962;
o Surface temperature at Port Erin Breakwater.
■ Bottom temperature at 10f. (Bay Fine)
+ Bottom temperature at 40f. (off Port Erin)

Maps 1-71

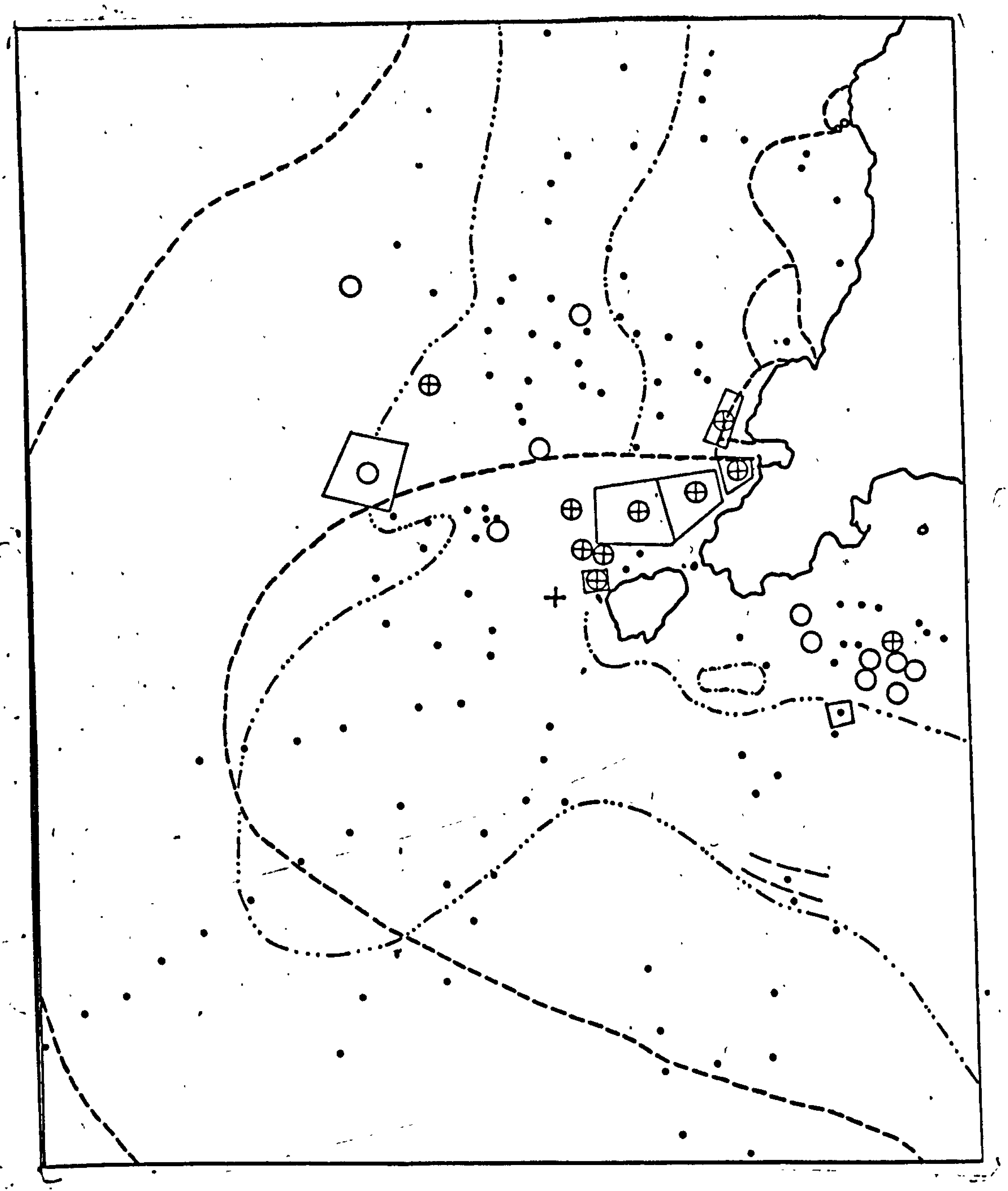


Map 1. Area investigated, bottom deposits and localities of dredging stations. ----- Approximate bounduaries of bottom deposits (after Jones 1951). — · — · 10 fathom, — · · —20 fathom, — ··· — 30 fathom.



Map. 2. Abundance of ectoprocts in the area sampled in terms of number of colonies per 1000 cm surface area of support (excluding erect zoophytes) suitable for ectoproct colonisation.

○ = less than 50 colonies,
◔ = 50 - 100 colonies,
◑ = 100 - 250 colonies,
◕ = 250 - 500 colonies,
● = more than 500 colonies per 1000 cm, rest as Map 1.

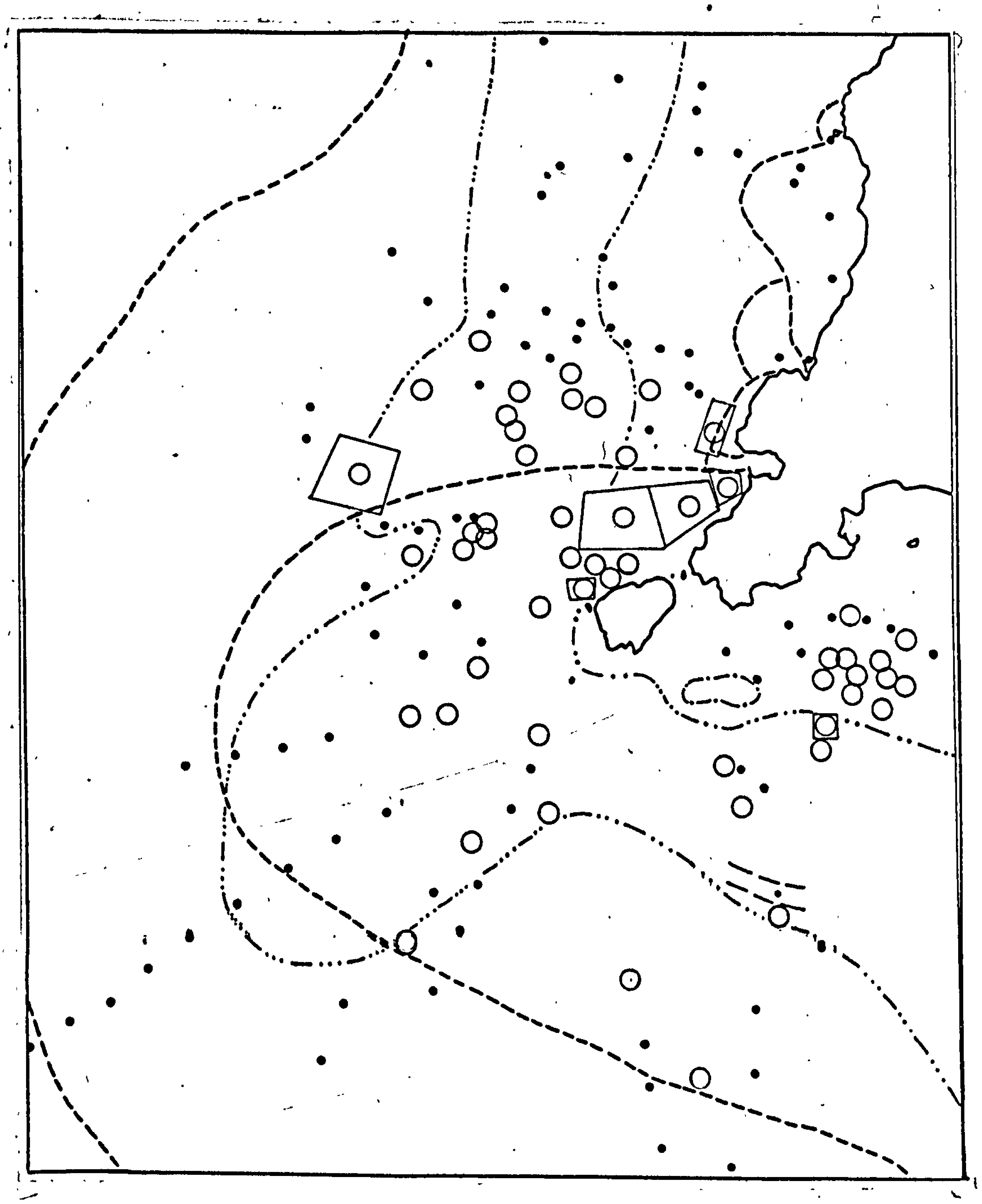


Map. 3. Crisidia cornuta and Crisia denticulata. Offshore distribution within the area sampled

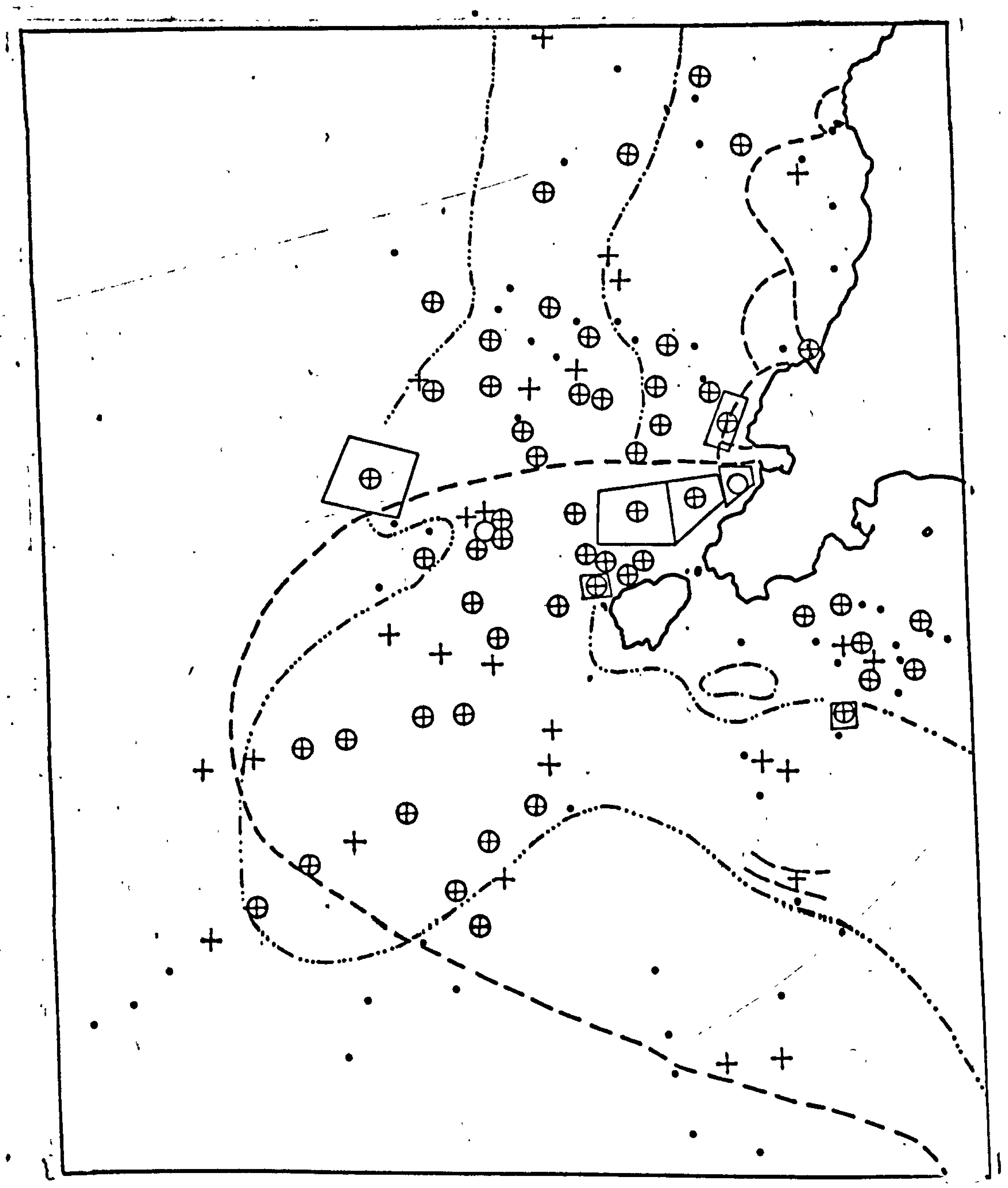
O = locality at which C.cornuta was obtained,

+ = locality at which C.denticulata was obtained,

• = locality at which neither was obtained, rest as Map 1.



Map. 4. Offshore distribution of Crisia eburnea in the area sampled.
O = locality at which C.eburnea was obtained,
• = locality at which C.eburnea was not obtained, rest as Map 1.

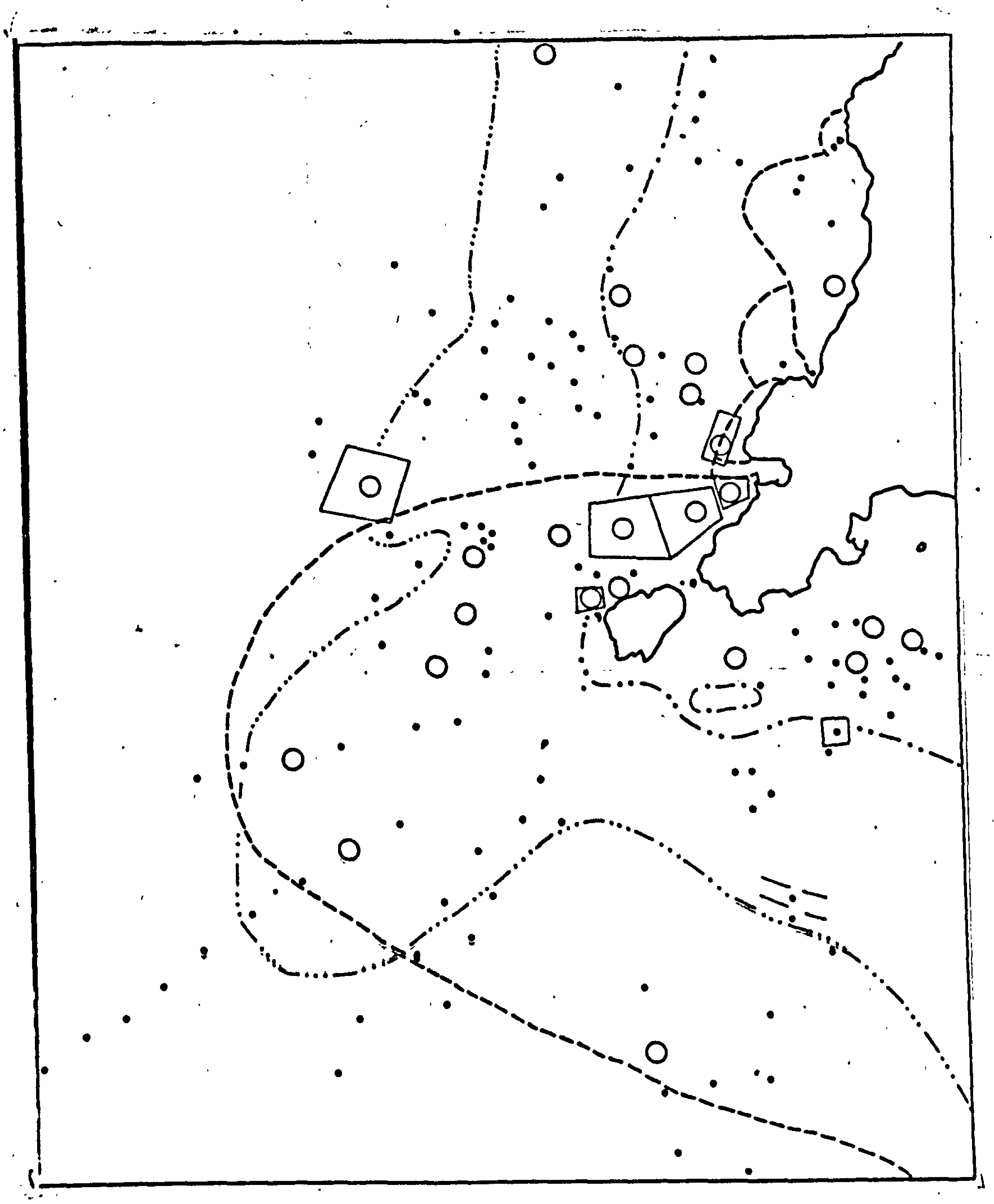


Map. 5. <u>Crisia aculeata</u> and <u>Crisia ramosa.</u> Offshore distribution within the area sampled.

+ = locality at which <u>C.aculeata</u> was obtained,

o = locality at which <u>C.ramosa</u> was obtained,

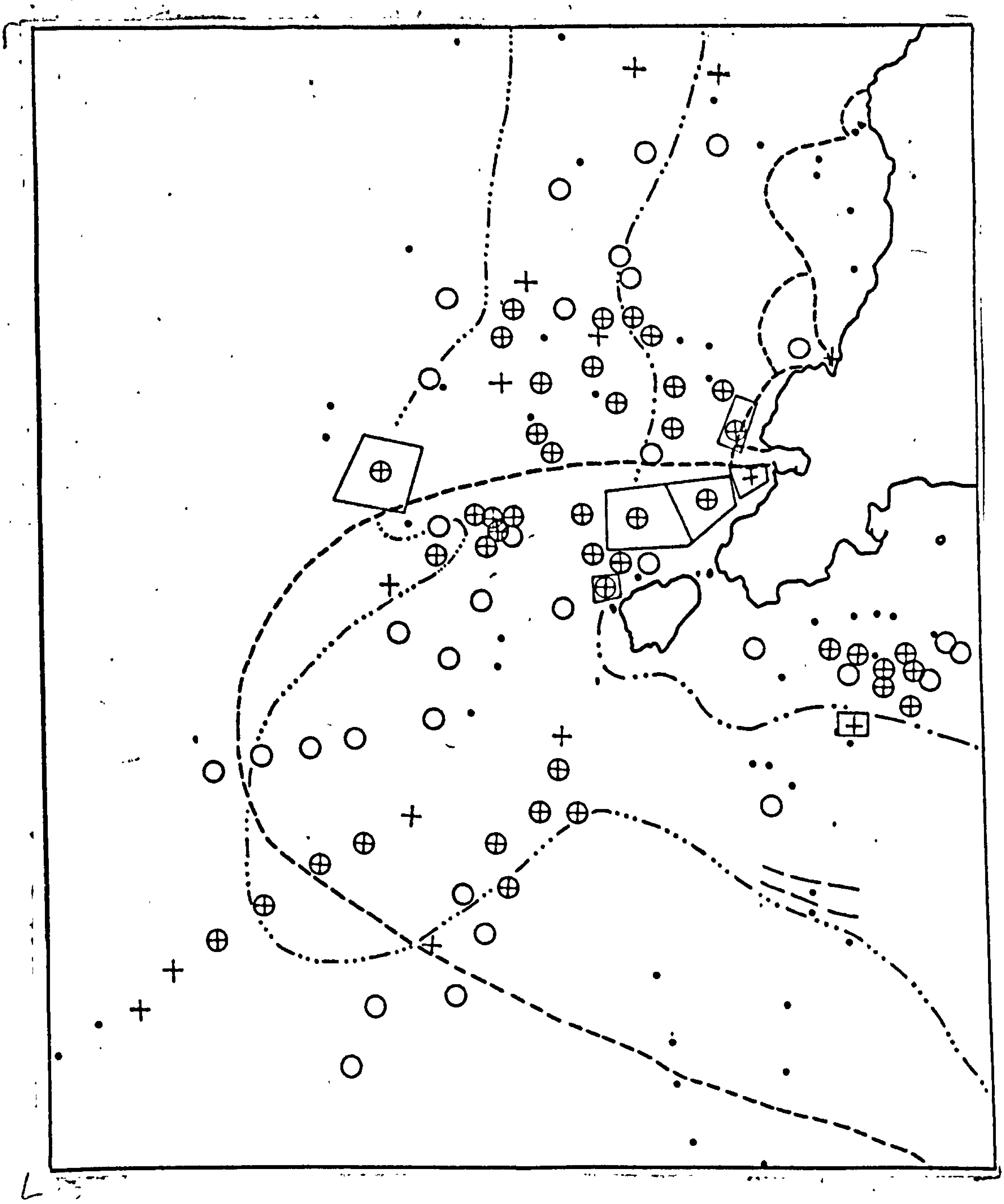
• = locality at which neither was obtained, rest as Map 1.



Map. 6. Tubulipora penicillata. Offshore distribution within the area sampled.

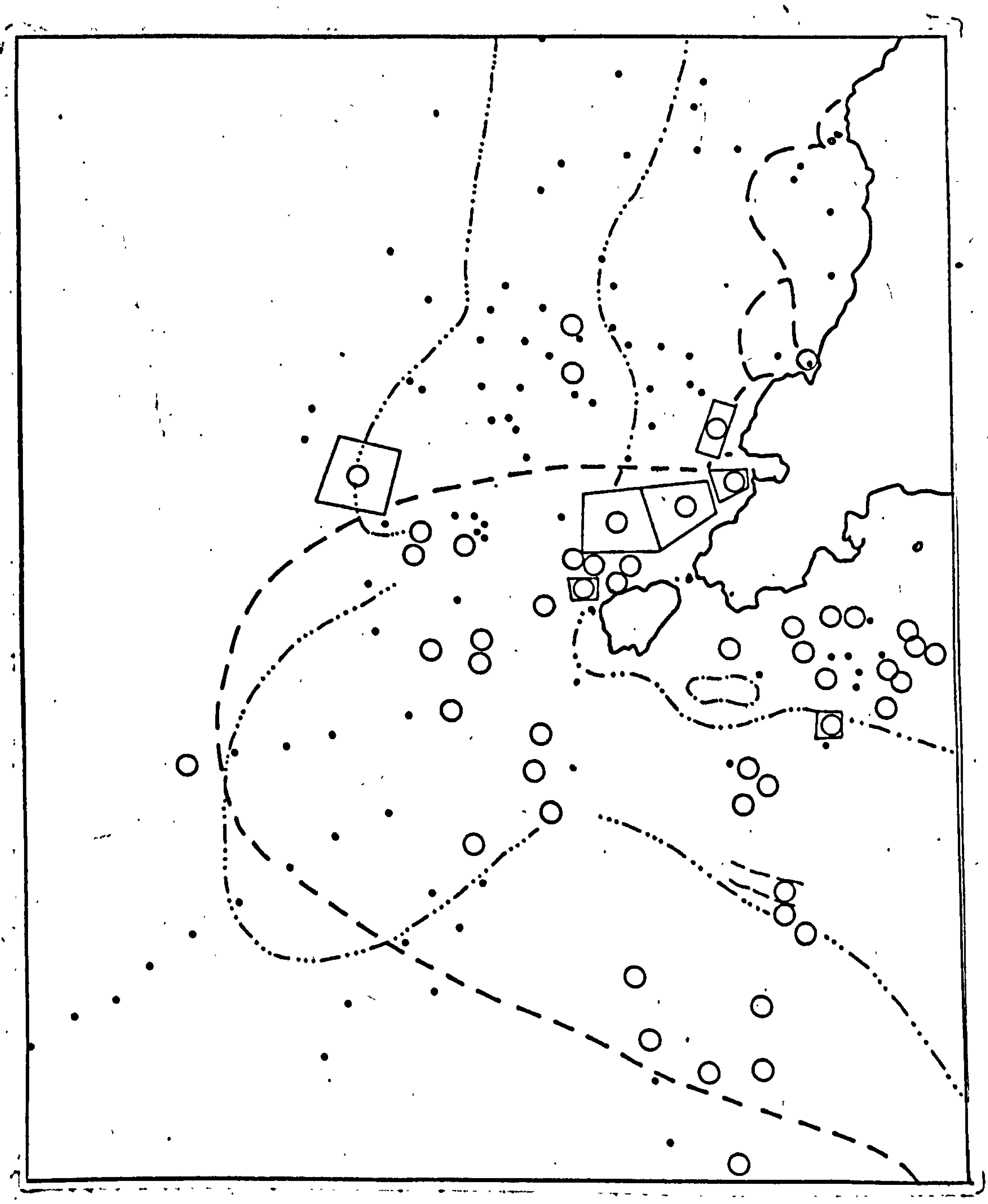
○ = locality at which T.penicillata was obtained,

• = locality at which T.penicillata was not obtained, rest as Map 1.

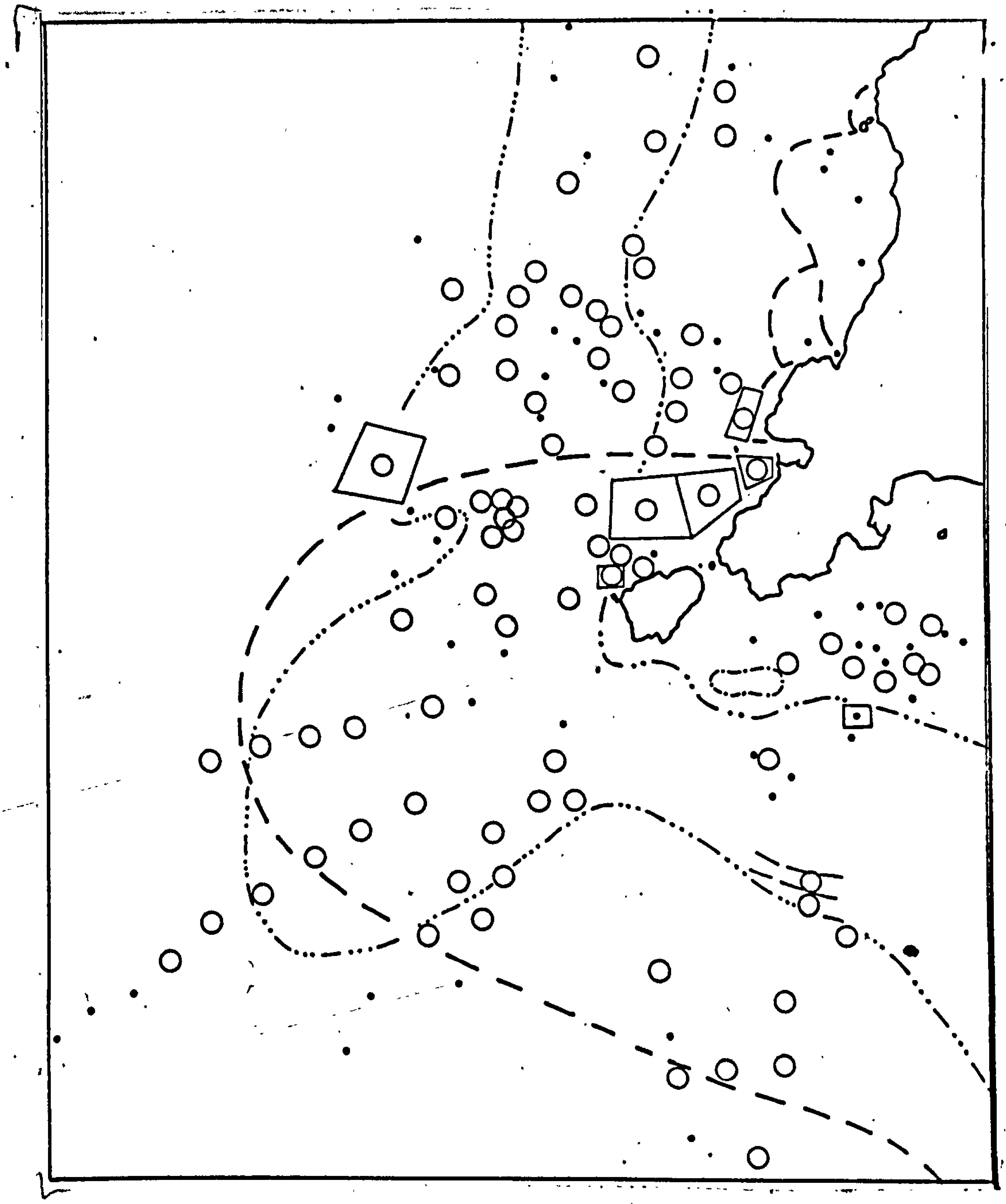


Map.7. Diaperoecia major and Diaperoecia johnstoni. Offshore distribution within the area sampled.

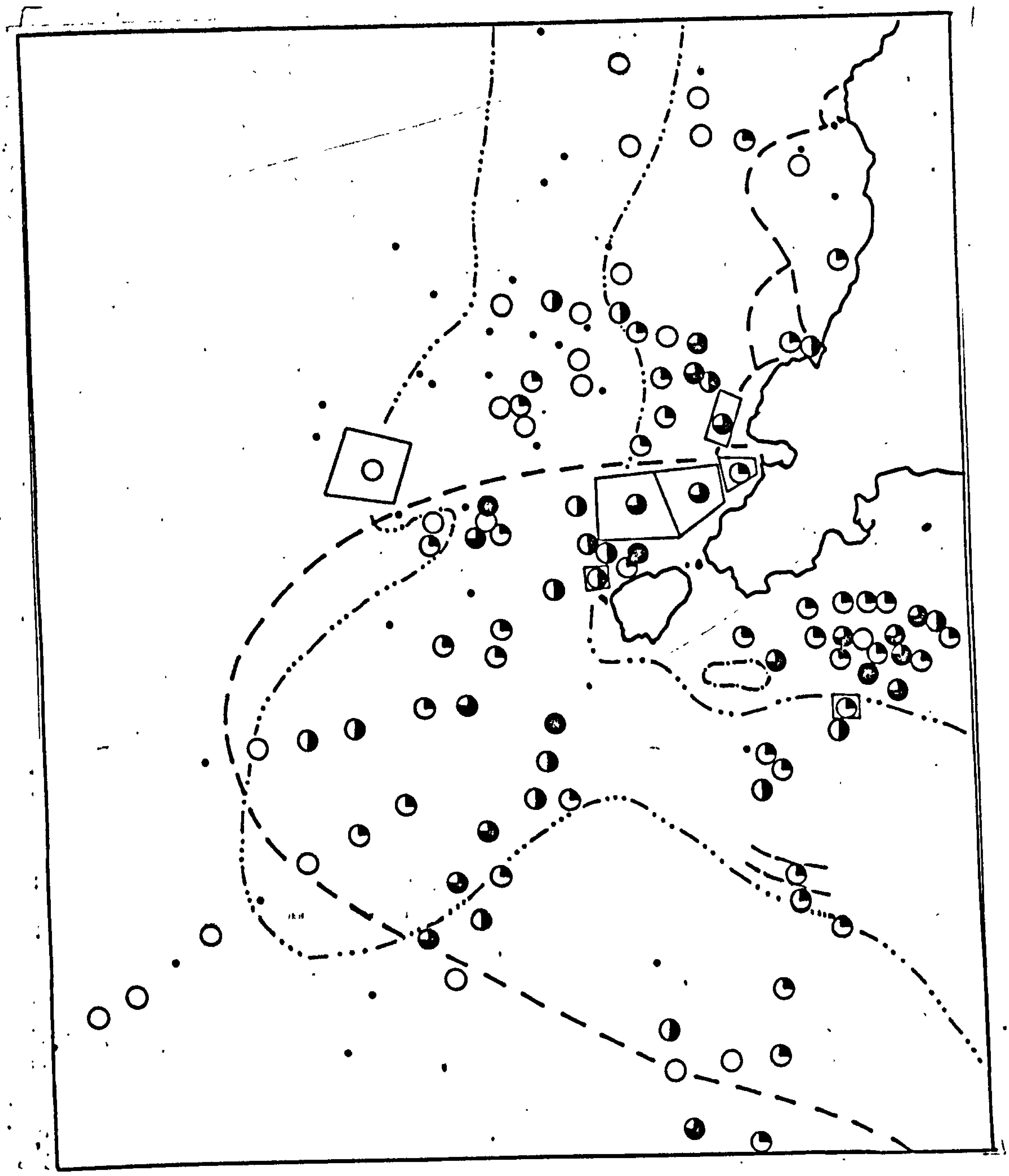
○ = locality at which D.major was obtained,
+ = locality at which D.johnstoni was obtained,
· = locality at which neither was obtained, rest as Map 1.



Map. 8. Diplosolen obelia. Offshore distribution within the area sampled.
O = locality at which D.obelia was obtained,
• = locality at which D.obelia was not obtained, rest as Map 1.

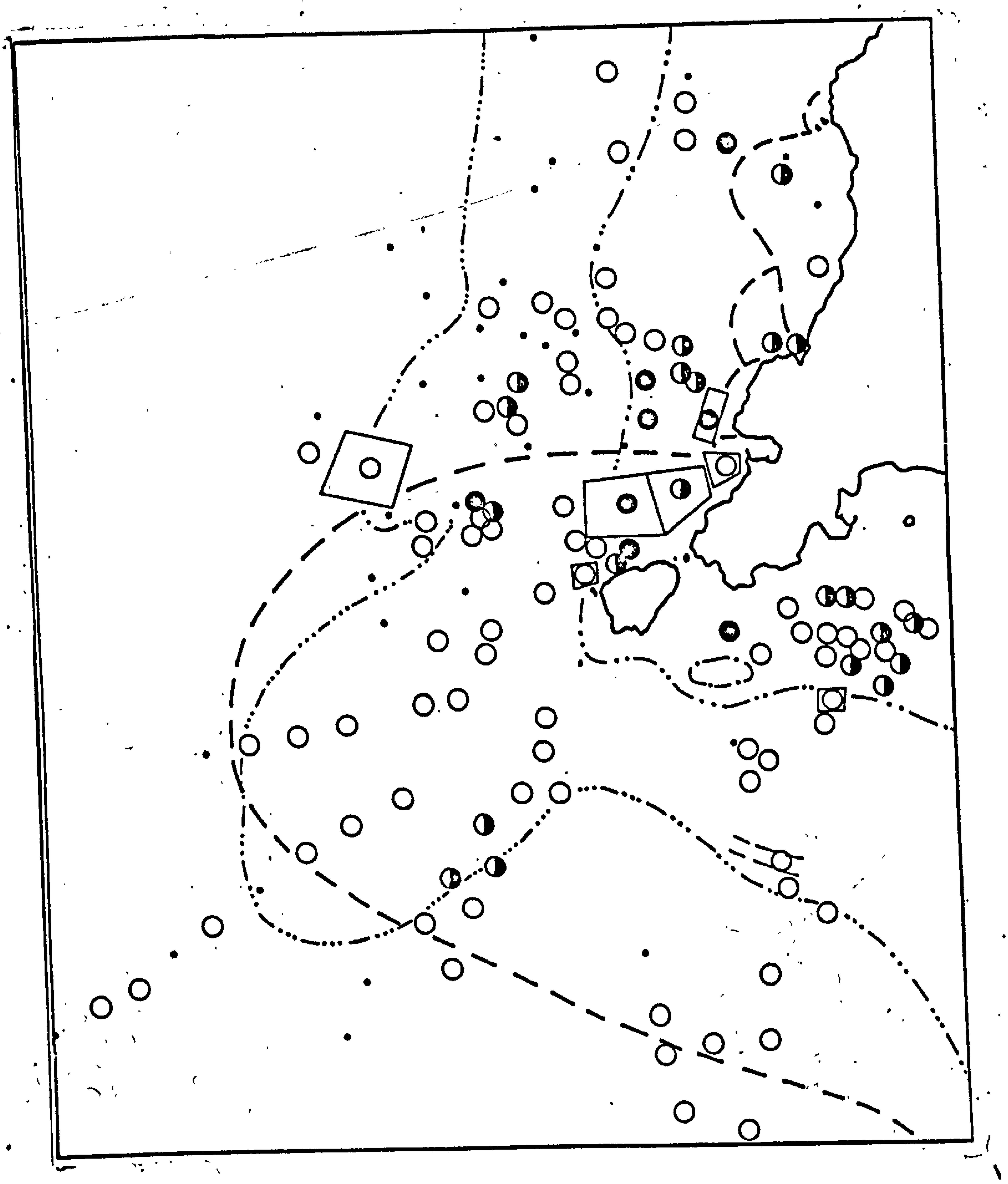


Map.9. Diastopora patina. Offshore distribution within the area sampled.
O = locality at which D.patina was obtained,
• = locality at which D.patina was not obtained, rest as Map 1.



Map.10. Diastopora suborbicularis. Offshore distribution and abundance within the area sampled in terms of no. of colonies per 1000 cm surface area of support (excluding erect zoophytes) suitable for ectoproct colonisation.

- • = locality at which D. suborbicularis was not obtained,
- ○ = less than 1 colony per 1000cm ,
- ◔ = 1 - 5 colonies per 1000cm ,
- ◑ = 5 - 10 colonies per 1000cm ,
- ◕ = 10 - 20 colonies per 1000cm ,
- ● = more than 20 colonies per 1000cm , rest as Map 1.



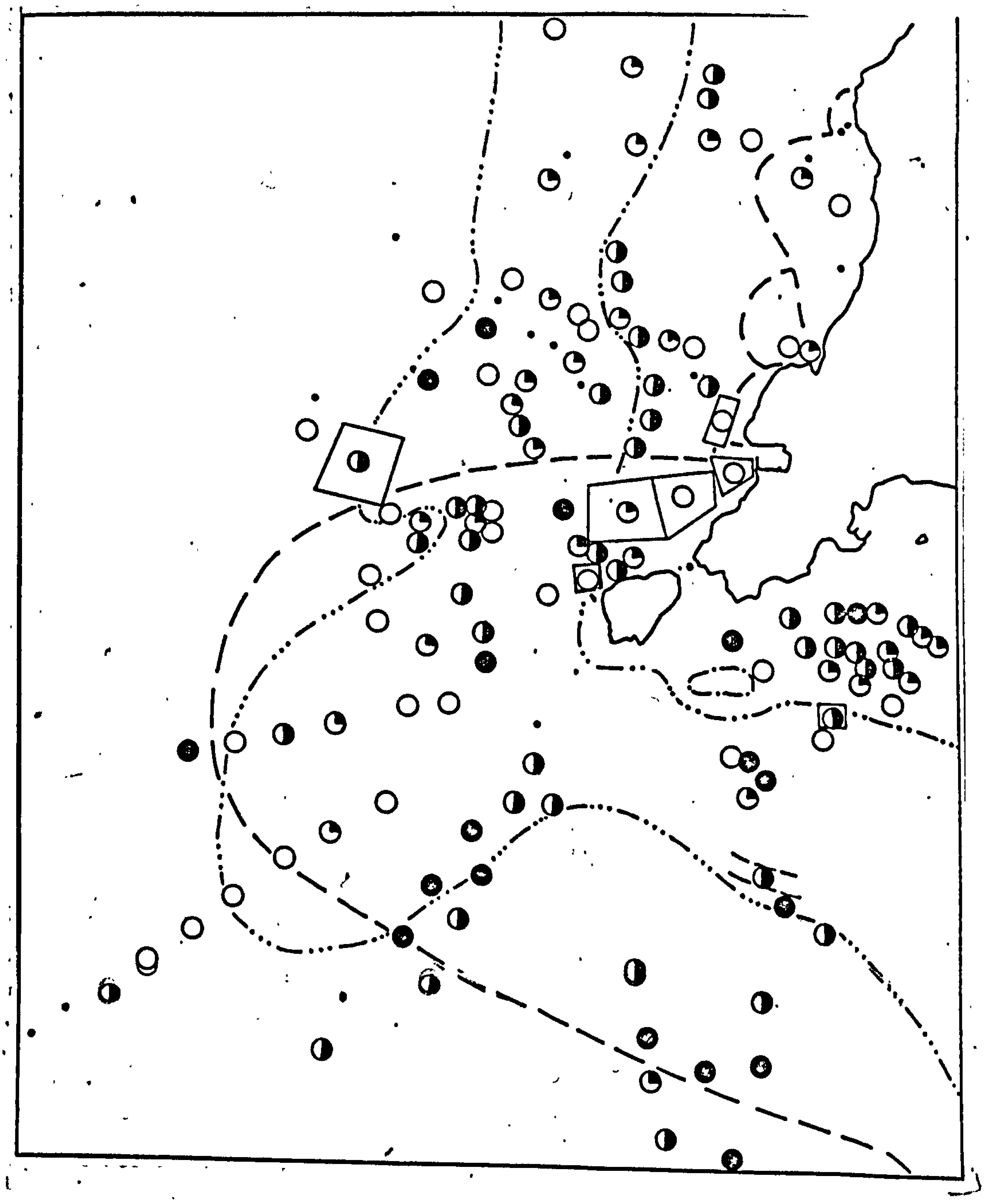
Map.11. Diastopora suborbicularis. Relative importance in the ectoproct population within the area sampled.

• = locality at which D. suborbicularis was not obtained,

○ = locality at which less than 5%,

◐ = locality at which from 5 - 10 %,

● = locality at which more than 10% of the ectoprocts are D. suborbicularis, rest as Map 1.



Map. 12. Lichenopora hispida. Offshore distribution and abundance within the area sampled in terms of no. of colonies per 1000 cm surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

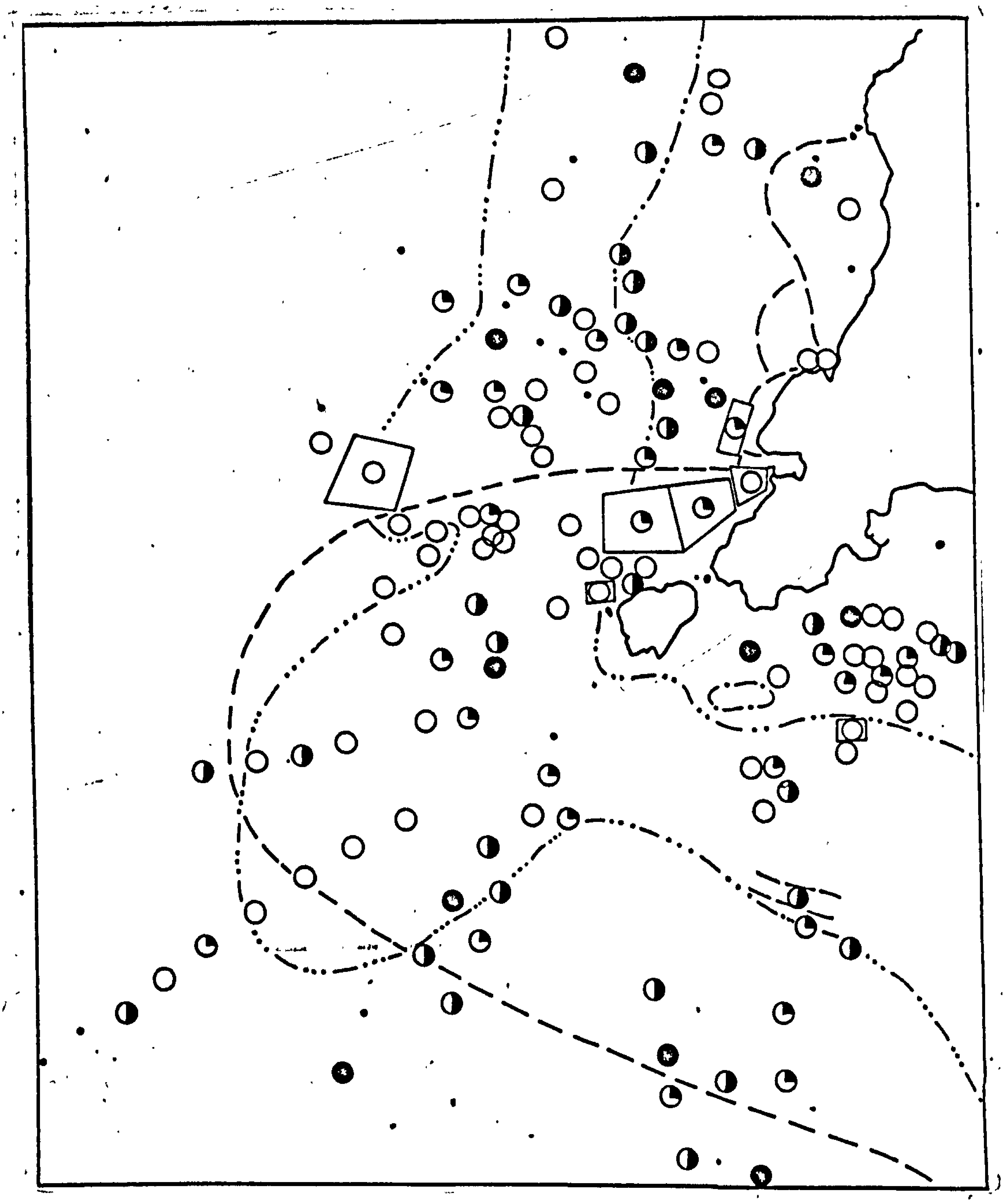
• = locality at which L.hispida was not obtained.

○ = less than 5 colonies per 1000cm ,

◔ = 5 -10 colonies per 1000cm ,

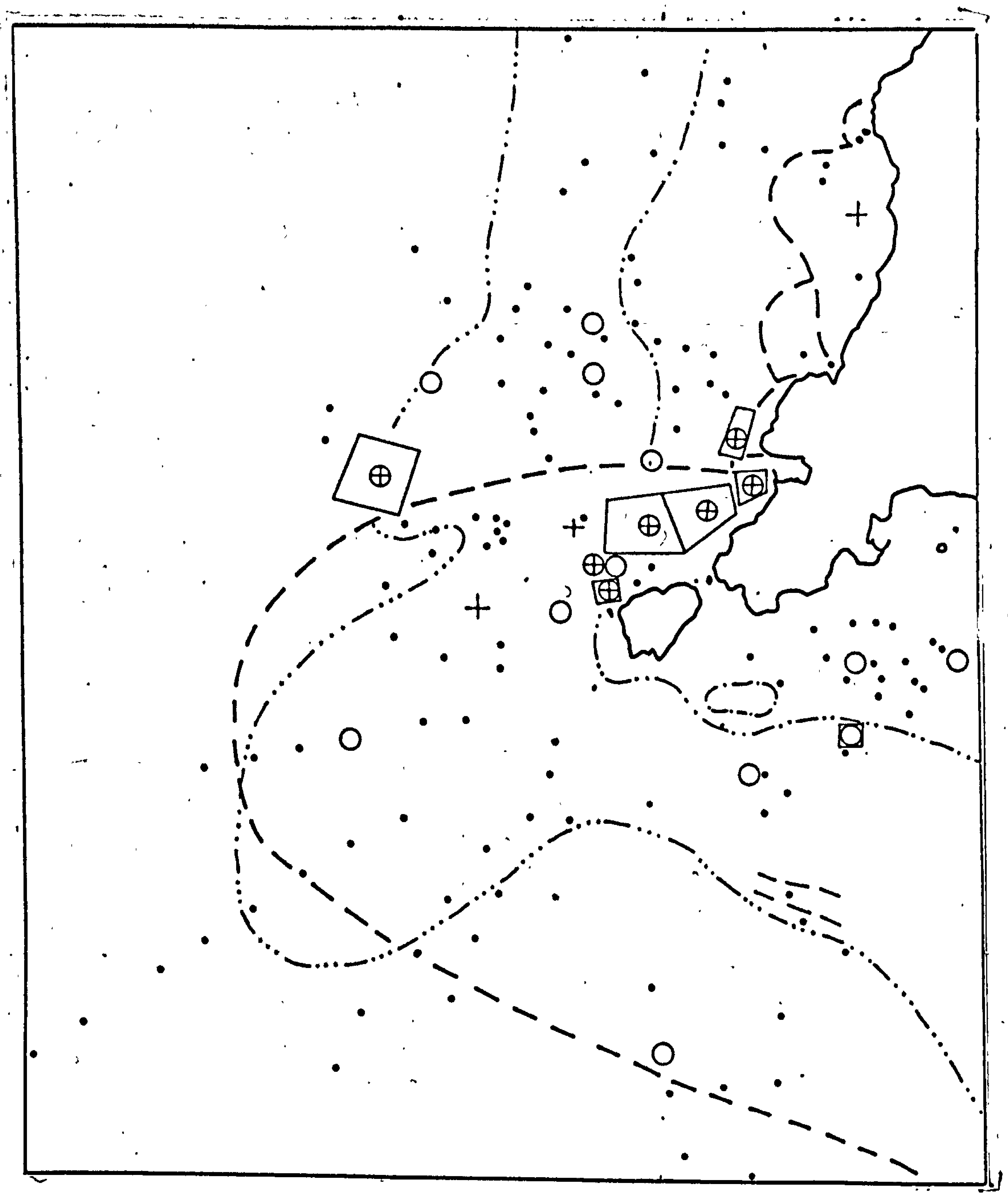
◑ = 10 - 20 colonies per 1000cm ,

● = more than 20 colonies per 1000cm , rest as Map 1.



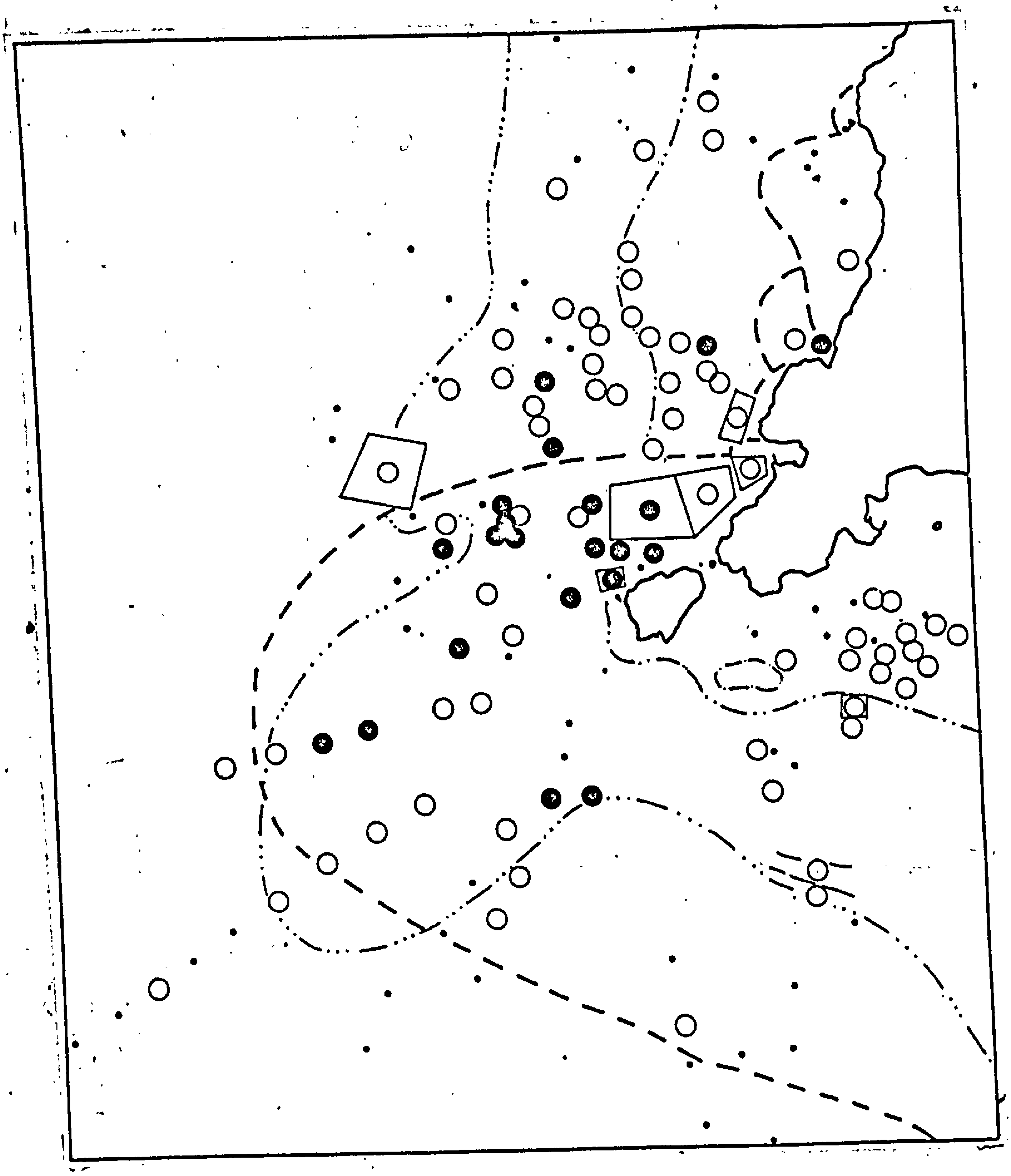
Map. 13. Lichenopora hispida. Relative importance in the ectoproct population within the area sampled.

• = locality at which L.hispida was not obtained
○ = locality at which less than 5% ,
◔ = locality at which from 5 - 10 %,
◑ = locality at which from 10 - 20 %,
● = locality at which more than 20 % of the ectoprocts are L. hispida, rest as Map 1.



Map. 14. Aetea anguinea and Aetea truncata. Distribution within the area sampled.

O = locality at which A.anguinea was obtained,
+ = locality at which A.truncata was obtained,
• = locality at which neither was obtained; rest as Map. 1.

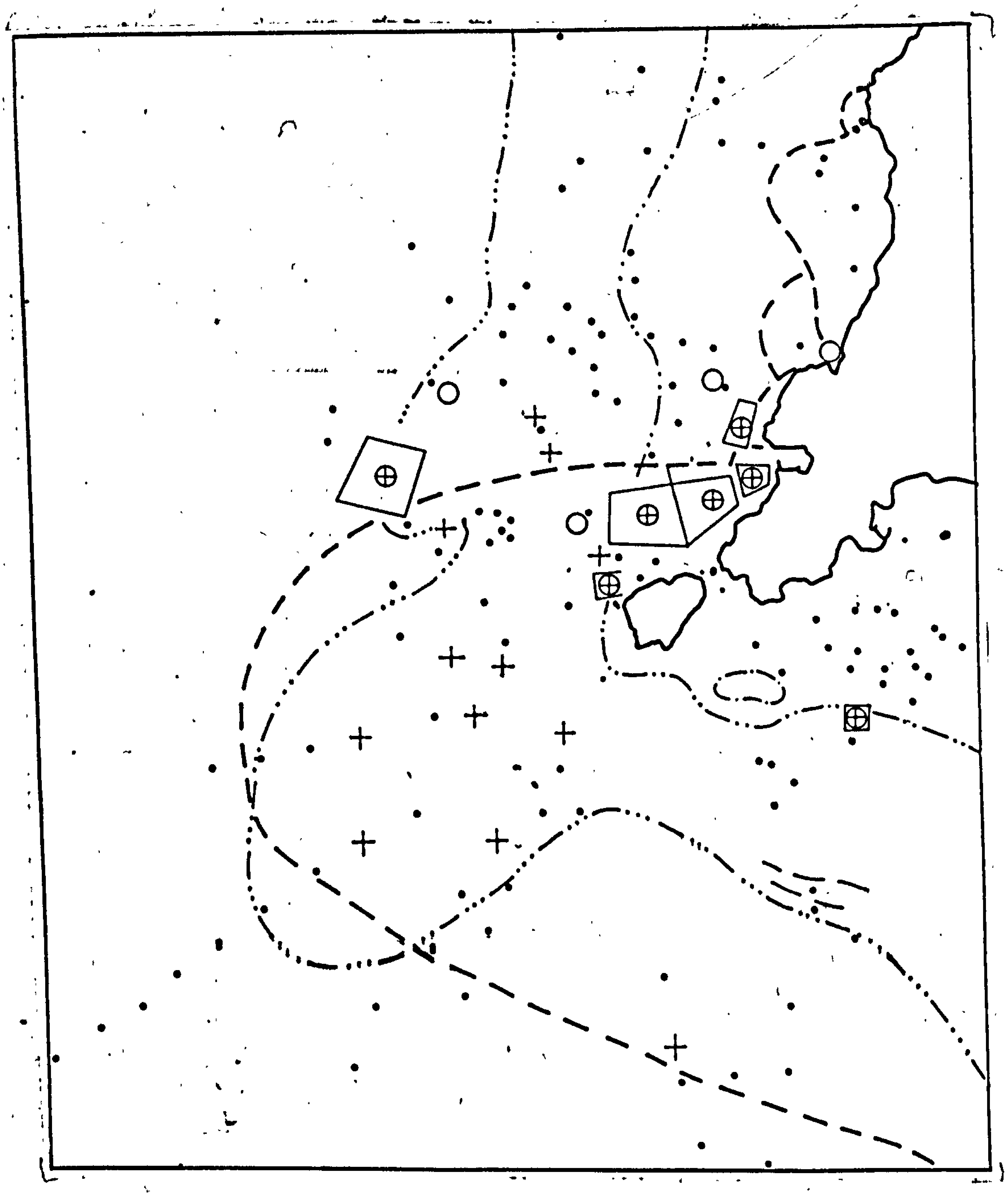


Map. 15. *Aetea sica*. Distribution and abundance within the area sampled in terms of number of colonies per 1000 cm surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

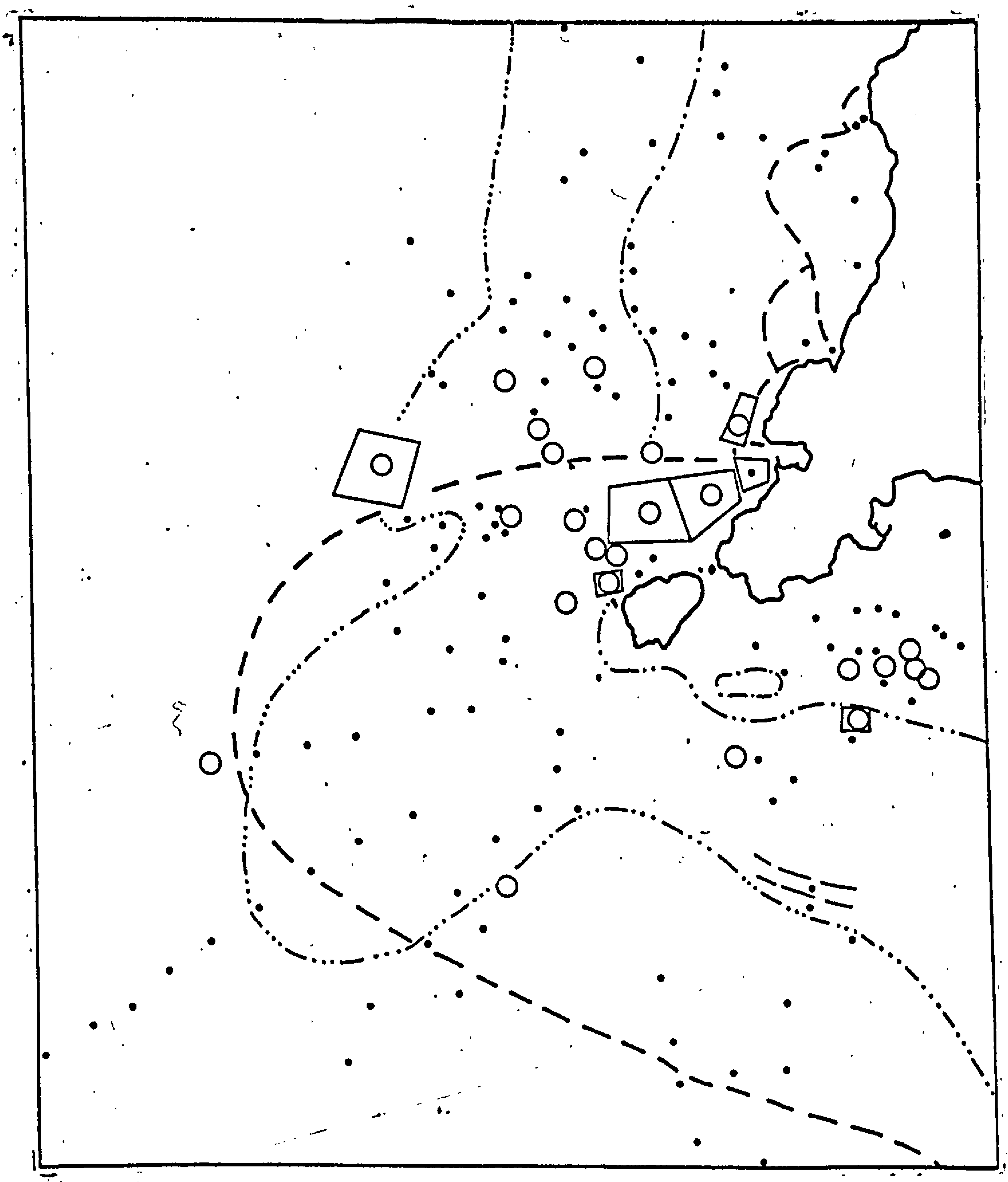
• = locality at which *A.sica* was not obtained,

O = less than 5 colonies per 1000cm ,

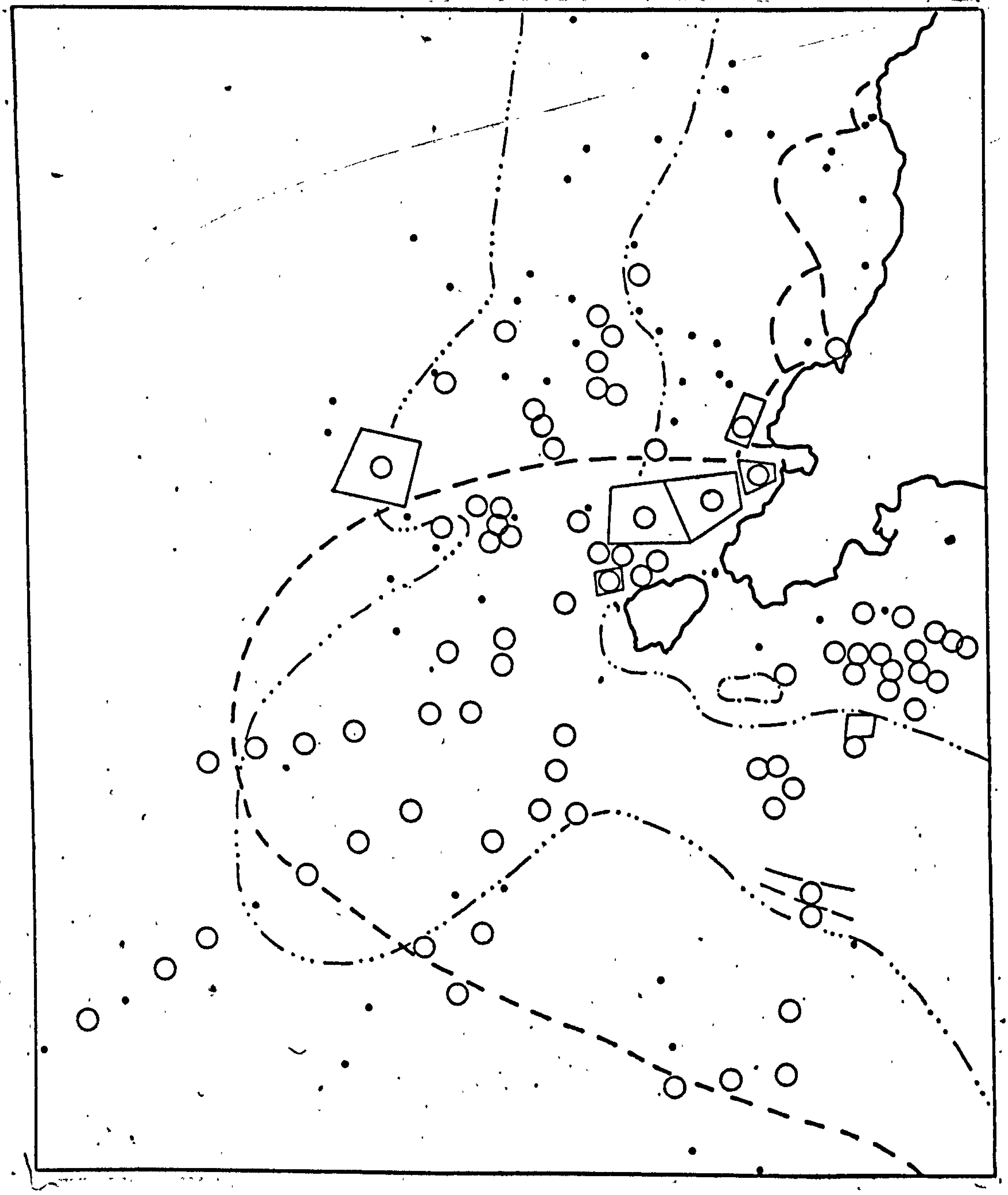
● = more than 5 colonies per 1000cm , rest as Map 1.



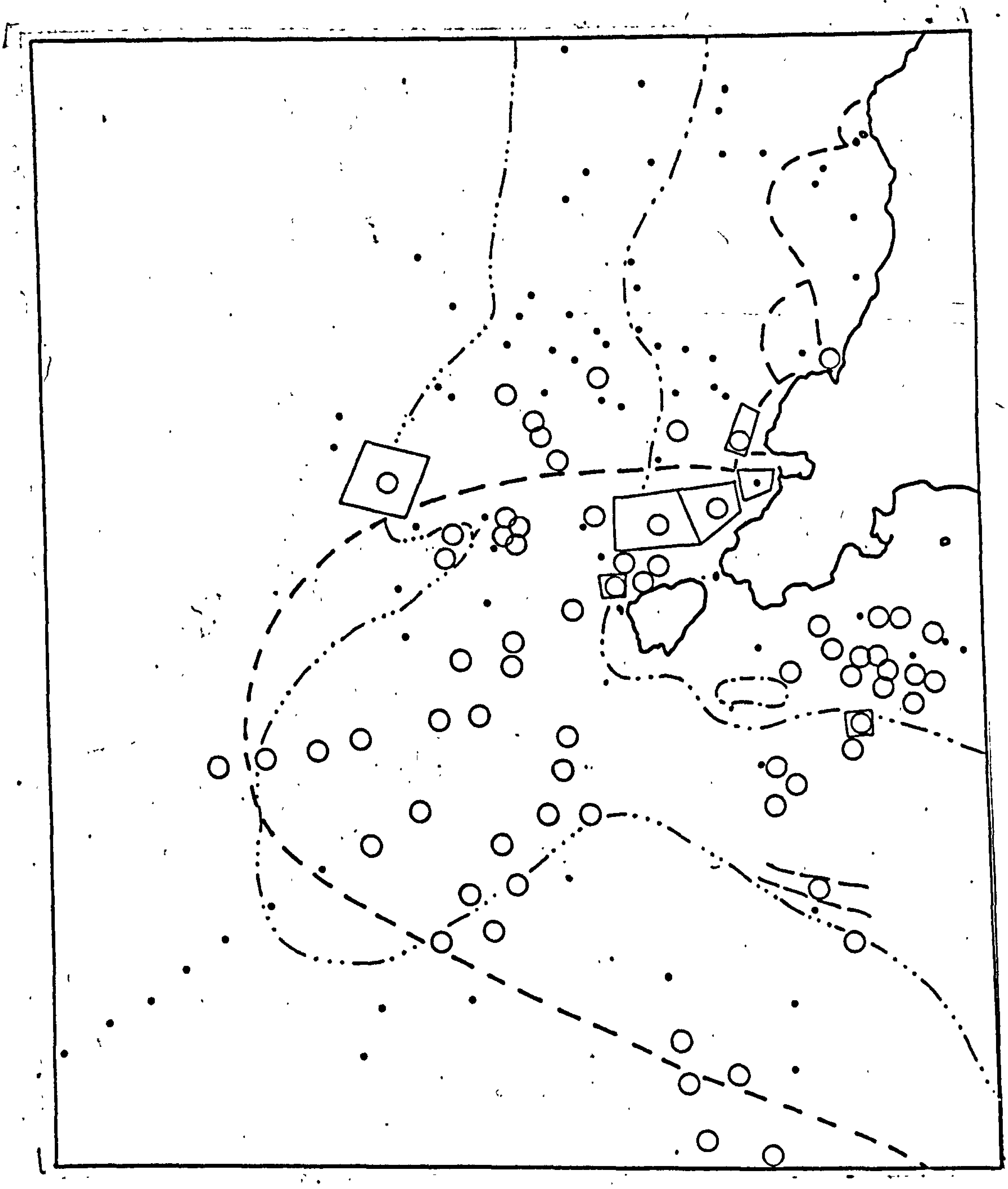
Map. 16. Scruparia chelata and Scruparia ambigua. Offshore distribution.
O = locality at which S.chelata was obtained;
+ = locality at which S.ambigua was obtained,
• = locality at which neither was obtained, rest as Map. 1.



Map 17. Eucratea loricata. Offshore distribution within the area sampled.
O = locality at which E. loricata was obtained,
• = locality at which E. loricata was not obtained, rest as Map 1.



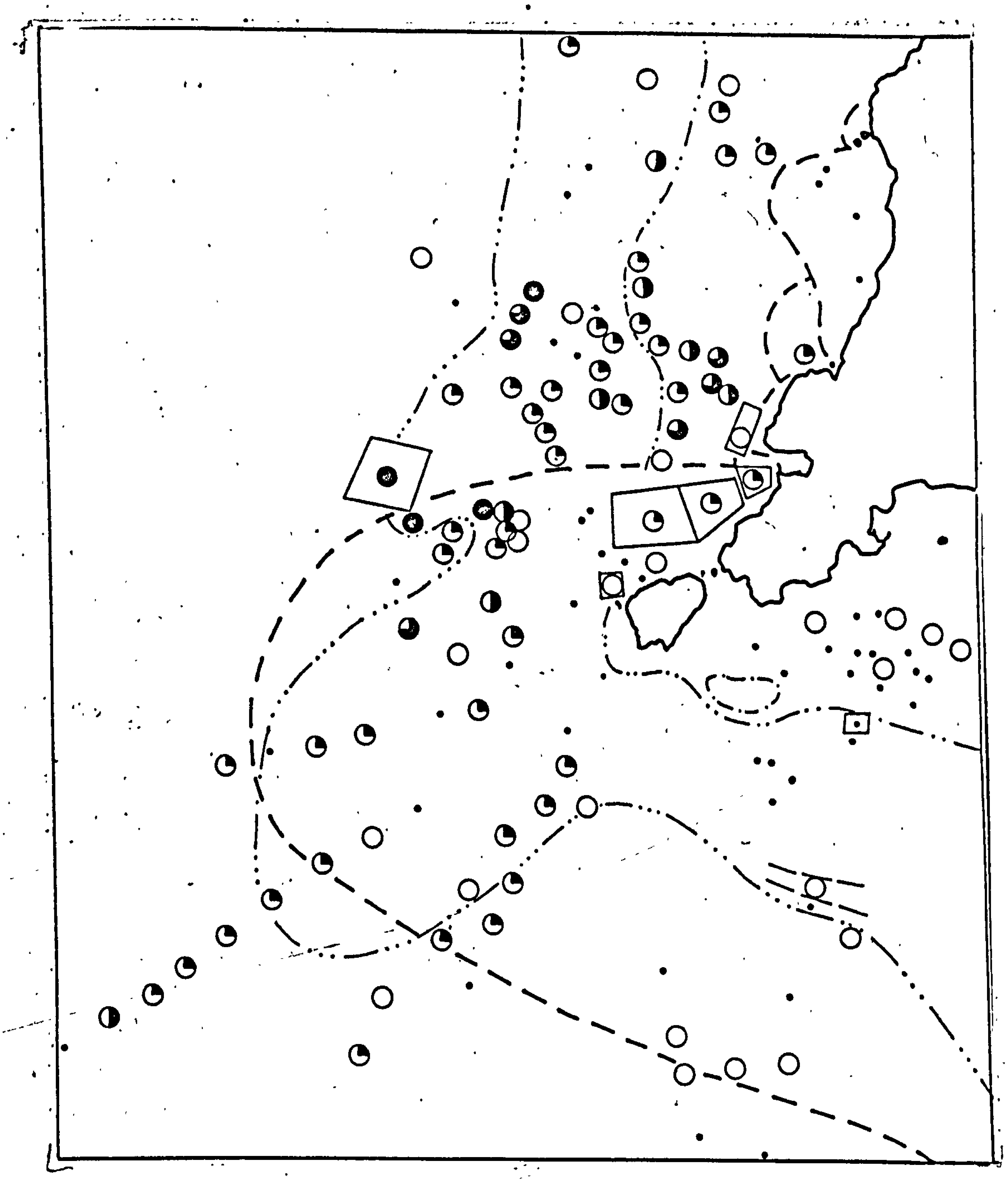
Map . 18. Electra pilosa. Offshore distribution within the area sampled.
O = locality at which E.pilosa was obtained,
• = locality at which E.pilosa was not obtained.



Map. 19. Pyripora catenularia. Offshore distribution within the area sampled.

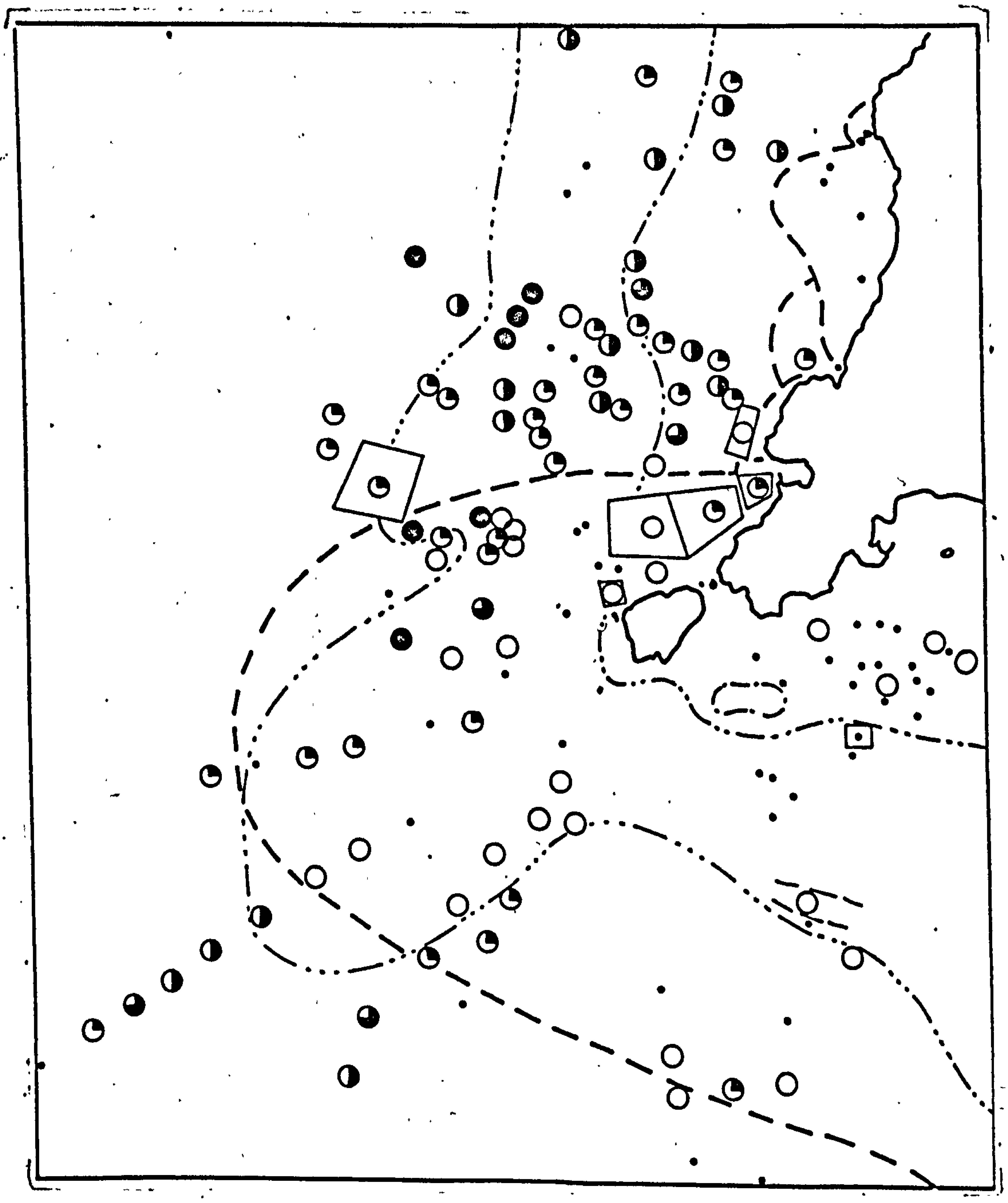
O = locality at which P.catenularia was obtained,

• = locality at which P.catenularia was not obtained, rest as Map.1.



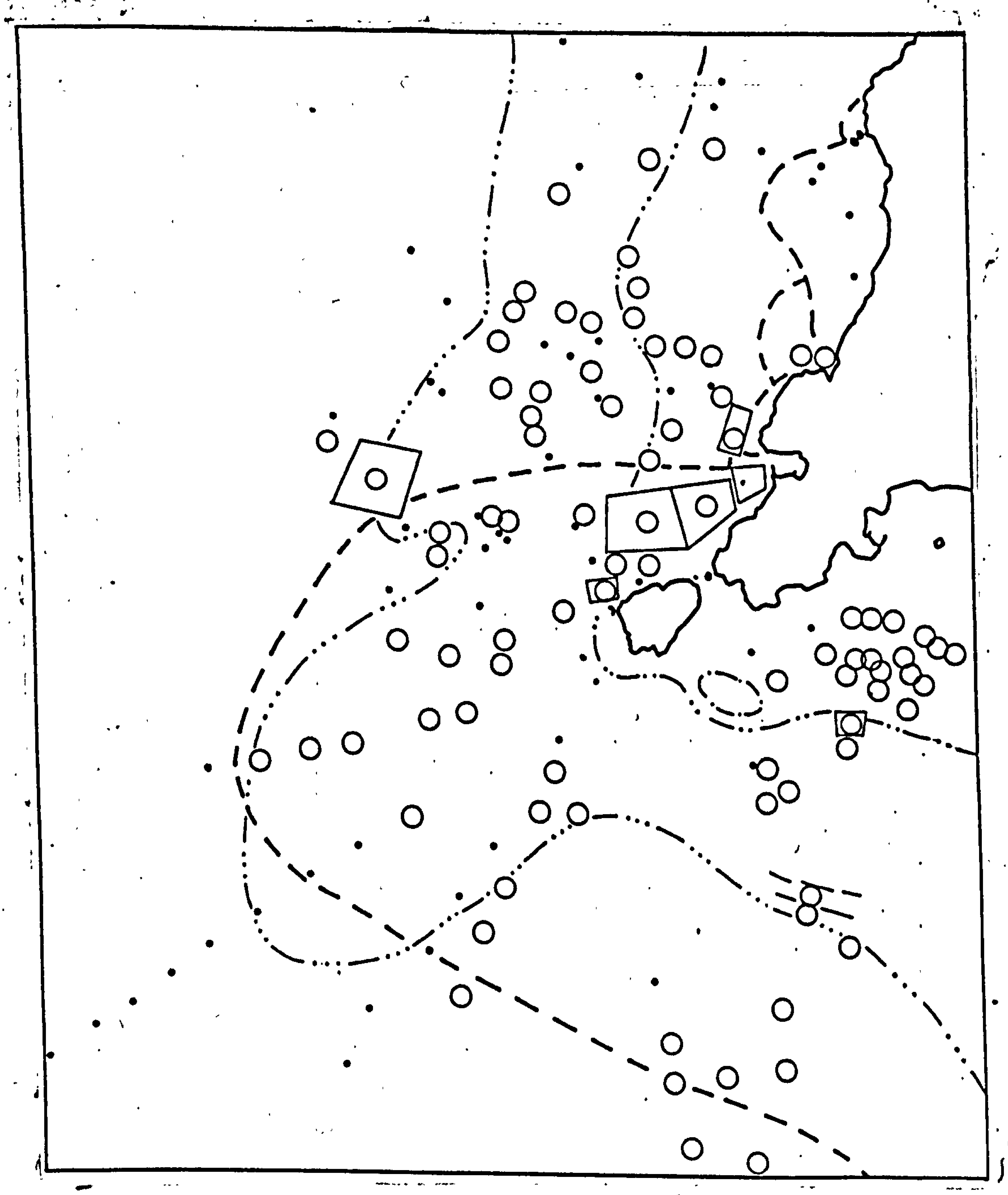
Map.20. Alderina imbellis. Distribution and abundance within the area sampled in terms of number of colonies per 1000cm surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

- • = locality at which A. imbellis was not obtained,
- ○ = less than 1 colony per 1000cm ,
- ◔ = 1 - 5 colonies per 1000cm ,
- ◑ = 5 -10 colonies per 1000cm ,
- ◕ = 110 -20 colonies per 1000cm ,
- ● = more than 20 colonies per 1000cm , rest as Map 1.



Map. 21. Alderina imbellis. Relative importance in the ectoproct population within the area sampled.

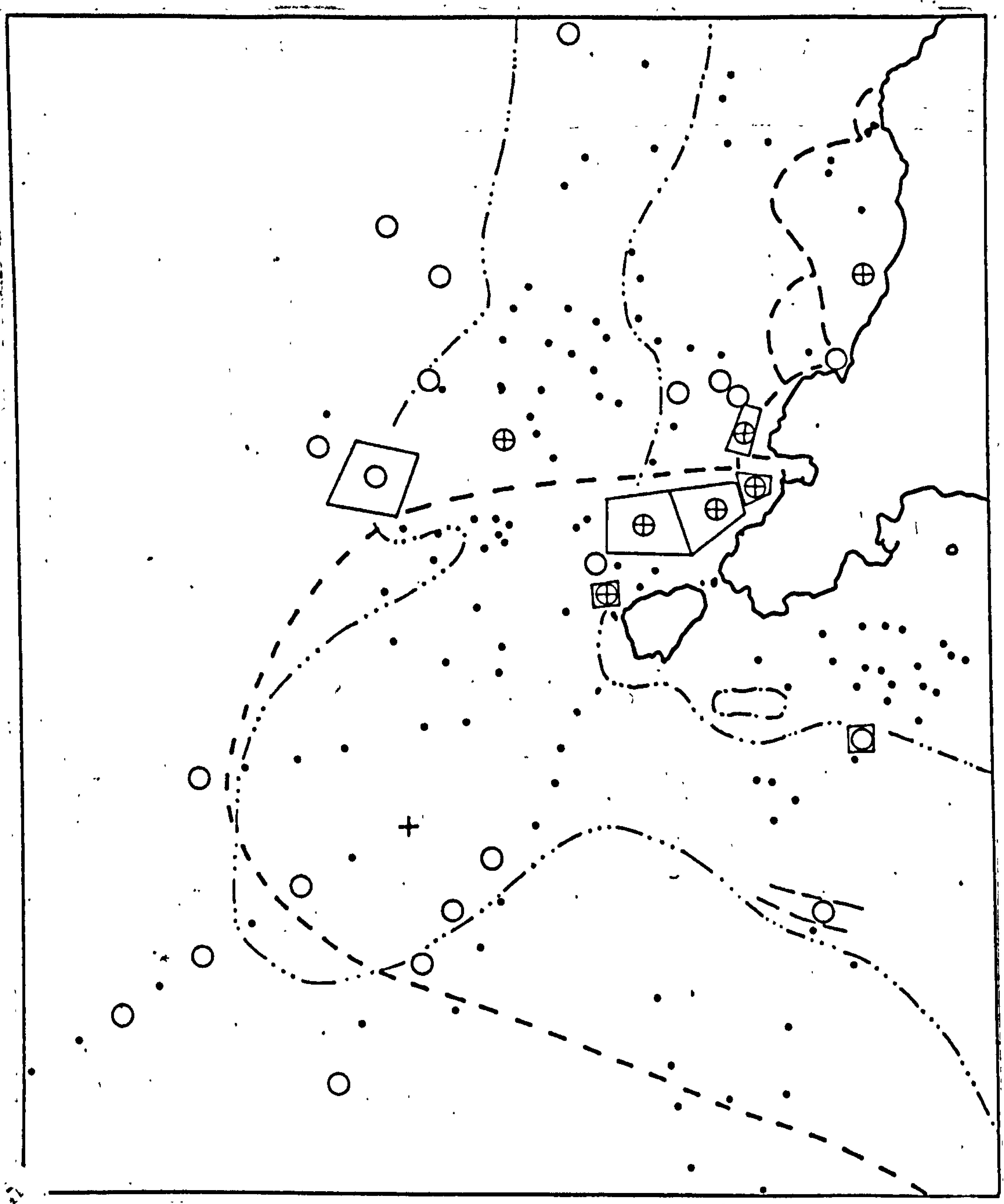
• = locality at which A.imbellis was not obtained,
○ = locality at which less than 1%,
◔ = locality at which from 1 - 5 %,
◑ = locality at which from 5 - 10 %,
◕ = locality at which from 10 - 20 %,
● = locality at which more than 20 % of the ectoprocts are A.imbellis, rest as Map.1.



Map. 22. Callopora lineata. Offshore distribution within the area sampled.

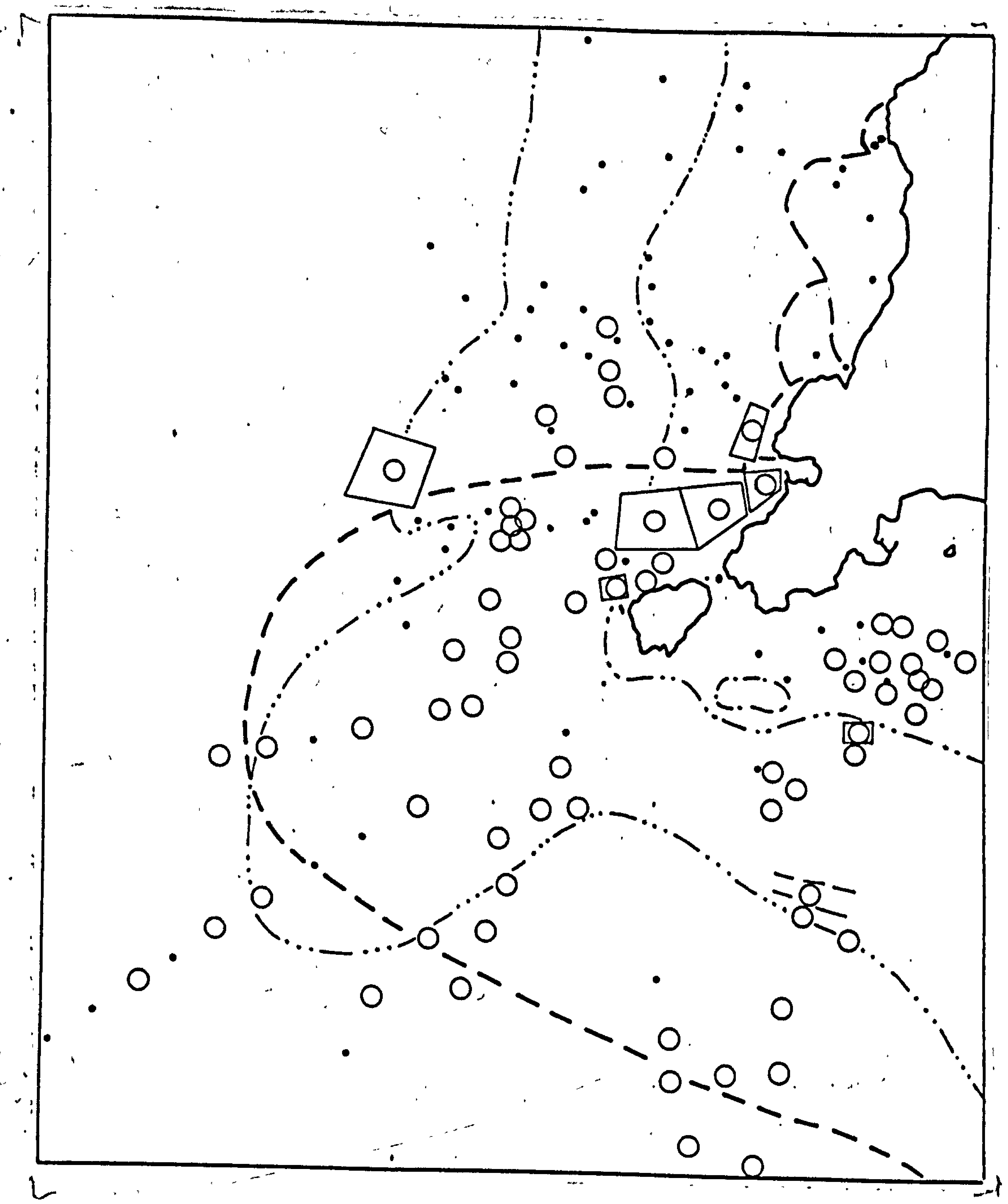
O = locality at which C.lineata was obtained.

• = locality at which C.lineata was not obtained, rest as Map.1.



Map .23. Callopora dumerili and Callopora craticula. Offshore distribution within the area sampled:

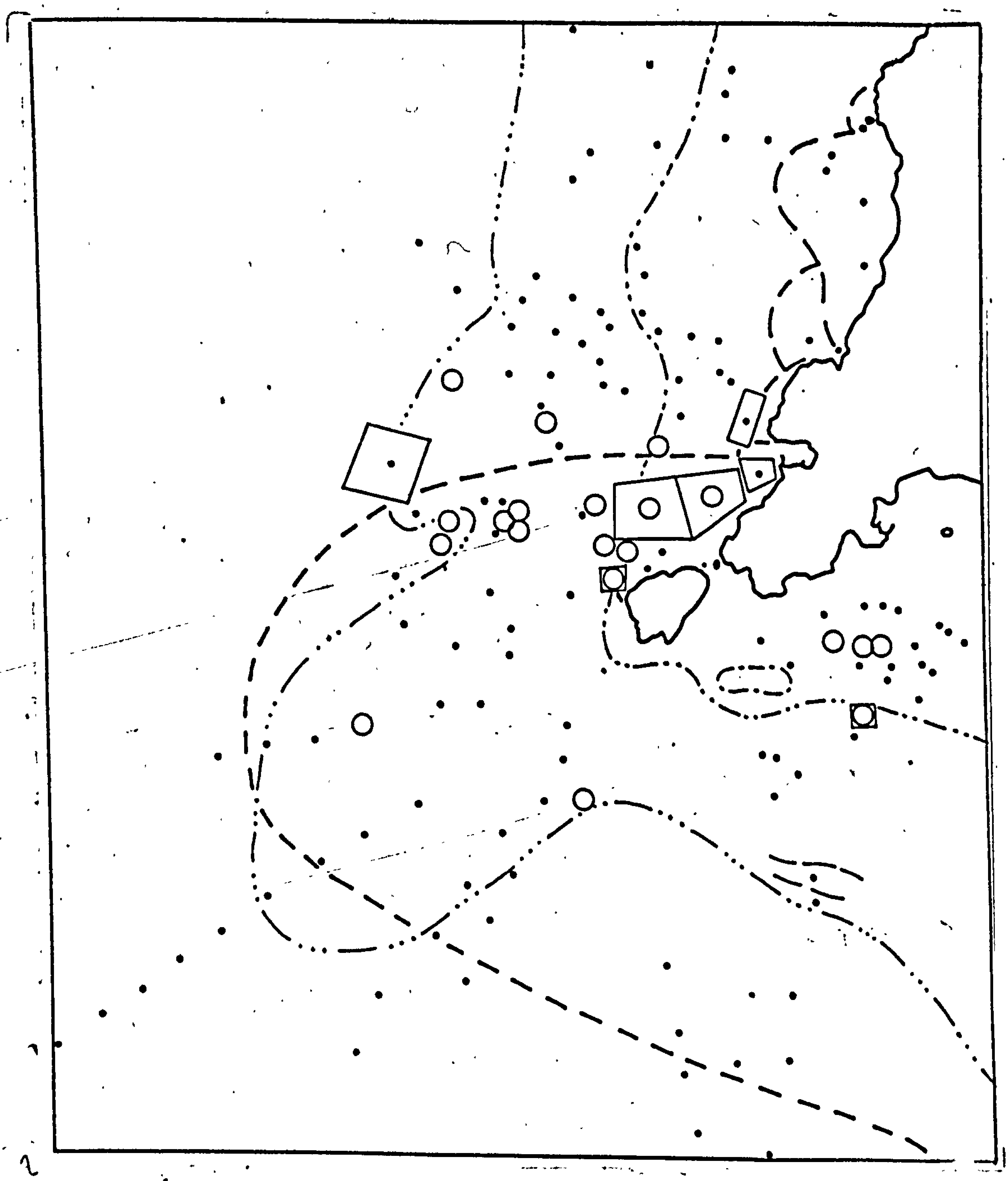
- O = locality at which C.dumerili was obtained,
- + = locality at which C.craticula was obtained,
- • = locality at which neither was obtained, rest as Map.1.



Map. 24. Amphiblestrum flemingi. Offshore distribution within the area sampled.

○ = locality at which A.flemingi was obtained,.

• = locality at which A.flemingi was not obtained, rest as Map.1.

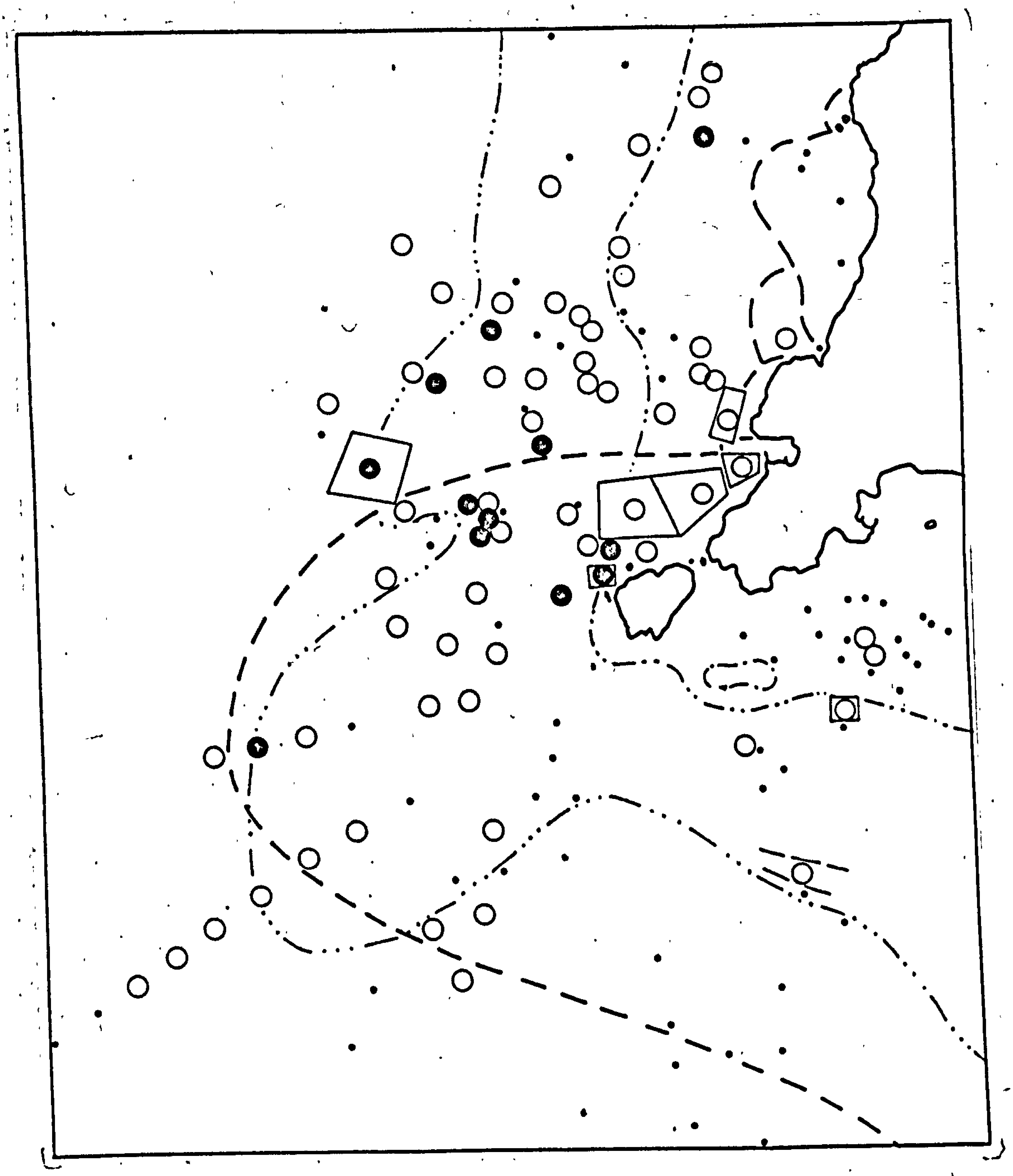


Map. 25. <u>Micropora</u> <u>coriacea</u>. Offshore distribution within the area sampled

O = locality at which M.coriacea was obtained,

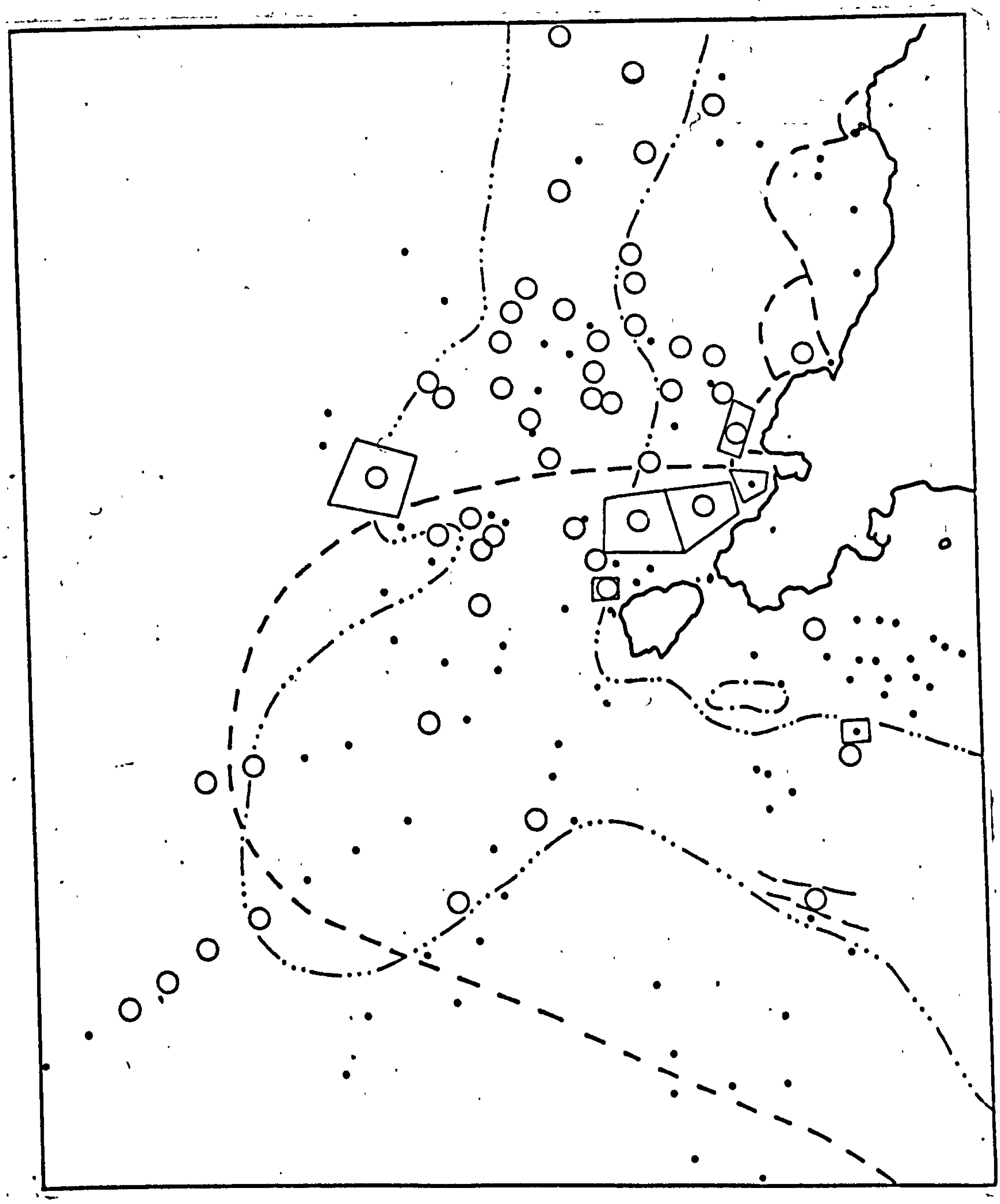
• = locality at which M.coriacea was not obtained,

rest as Map 1.

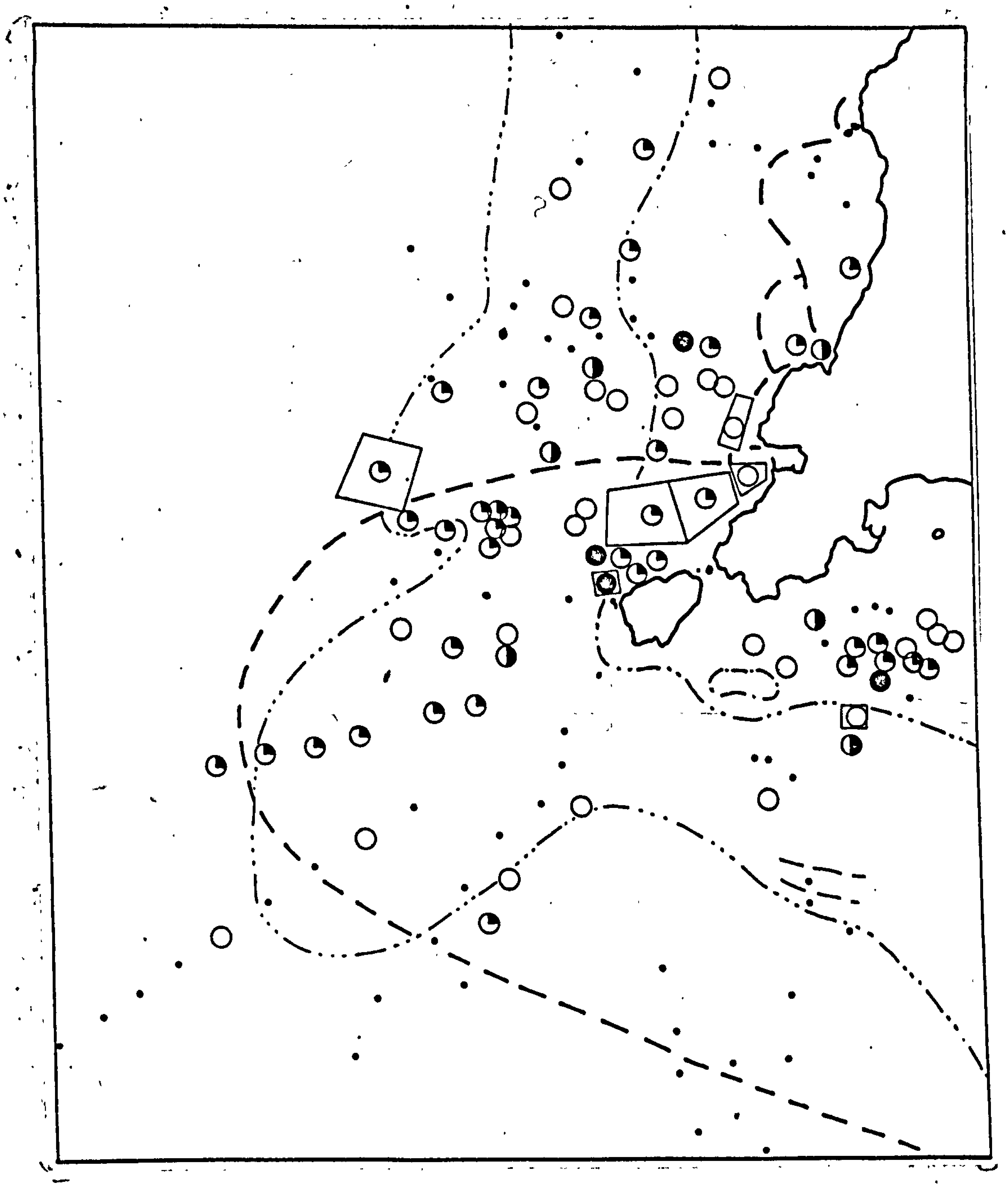


Map. 26. Cellaria fistulosa. Distribution and abundance within the area sampled in terms of number of colonies per 1000 cm² surface area suitable for ectoproct colonization.

• = locality at which C. fistulosa was not obtained,
O = less than 5 colonies per 1000 cm²,
● = more than 5 colonies per 1000 cm², rest as Map. 1.

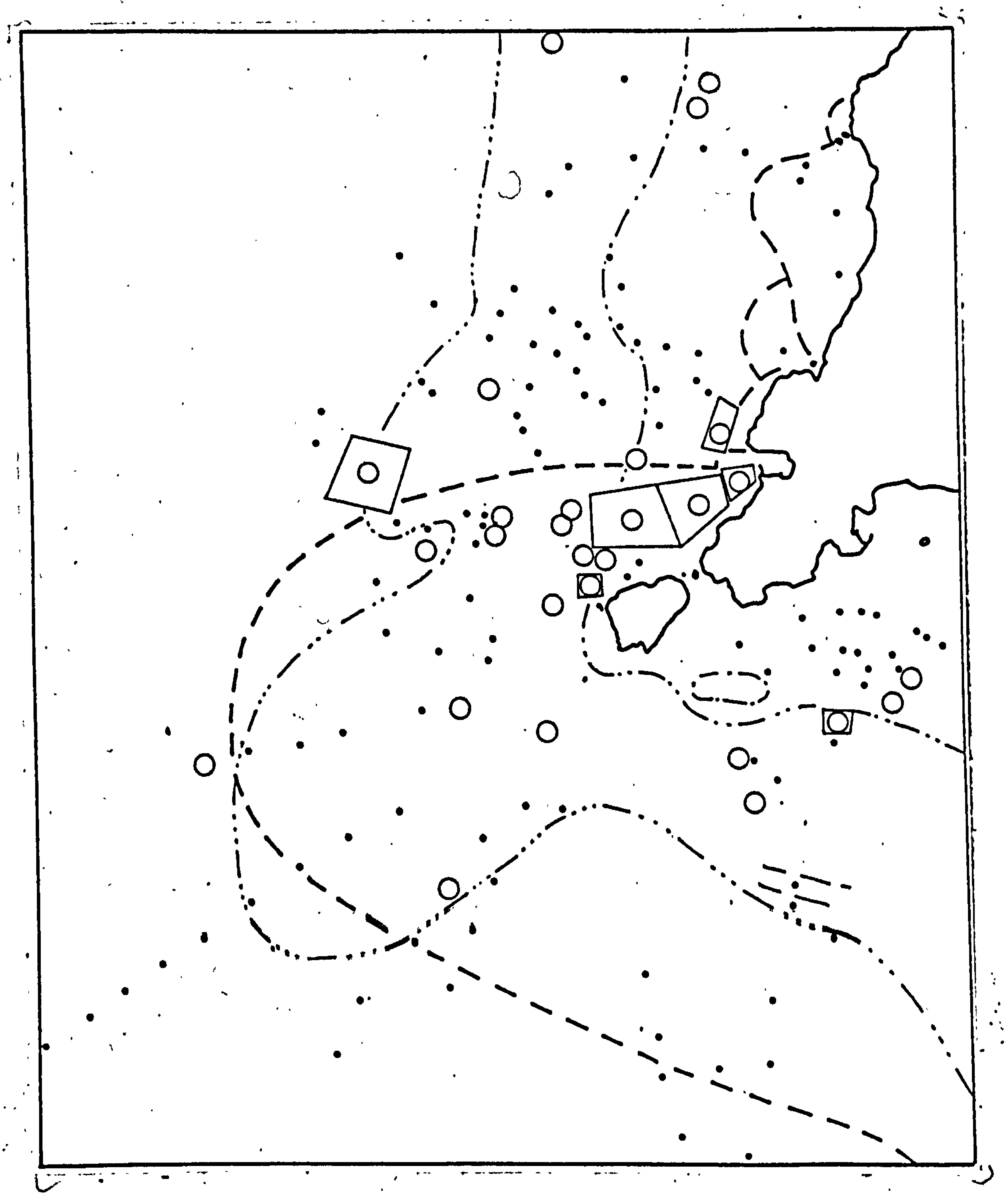


Map. 27. Cellaria sinuosa. Offshore distribution within the area sampled.
O = locality at which C.sinuosa was obtained,
• = locality at which C.sinuosa was not obtained, rest as Map.1.

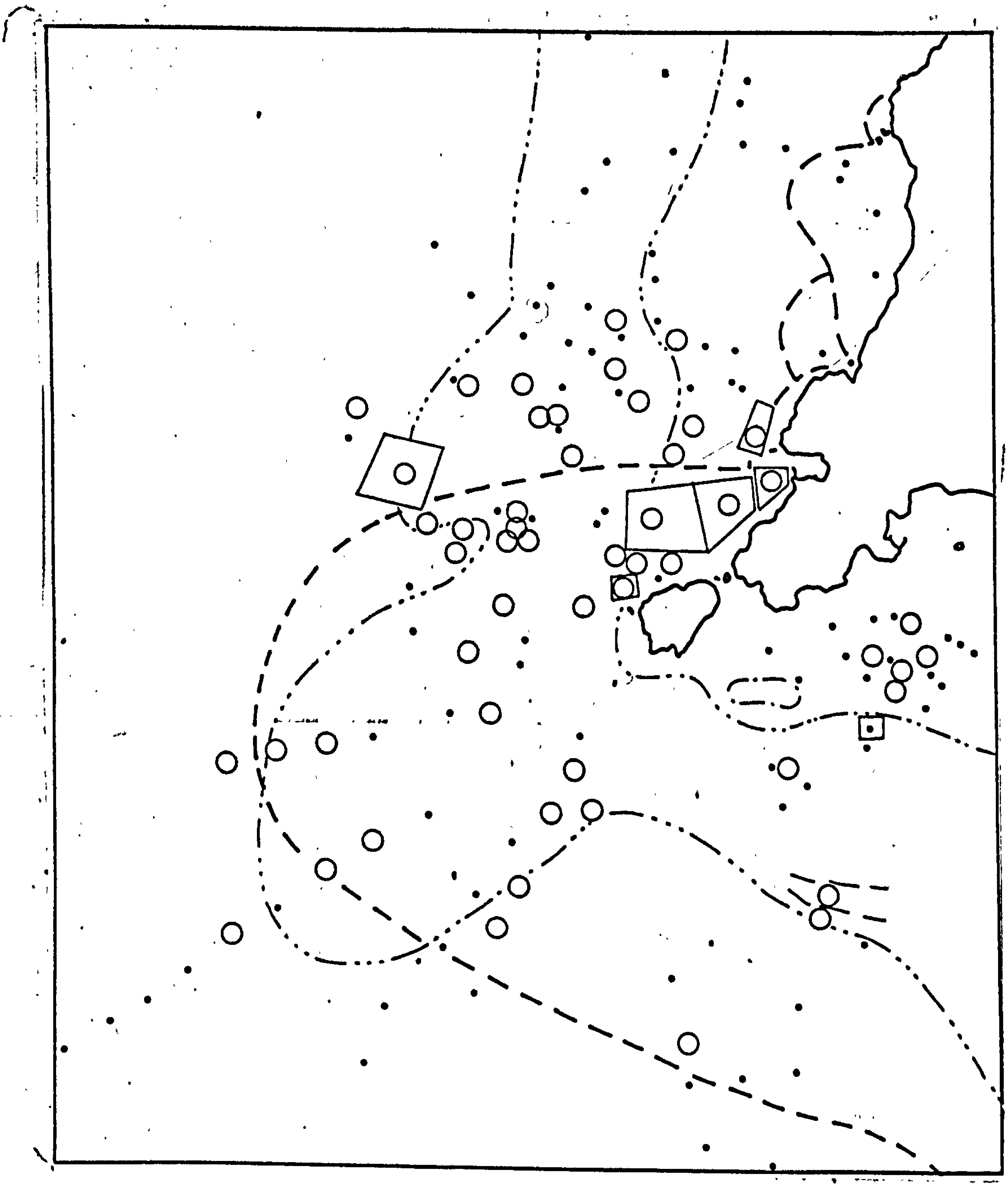


Map. 28. Scrupocellaria scruposa. Offshore distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

• = locality at which S. scruposa was not obtained
○ = less than 1 colony per 1000cm^2,
◔ = 1 - 5 colonies per 1000 cm^2,
◑ = 5 - 10 colonies per 1000 cm^2,
● = more than 10 colonies per 1000 cm^2, rest as Map 1.



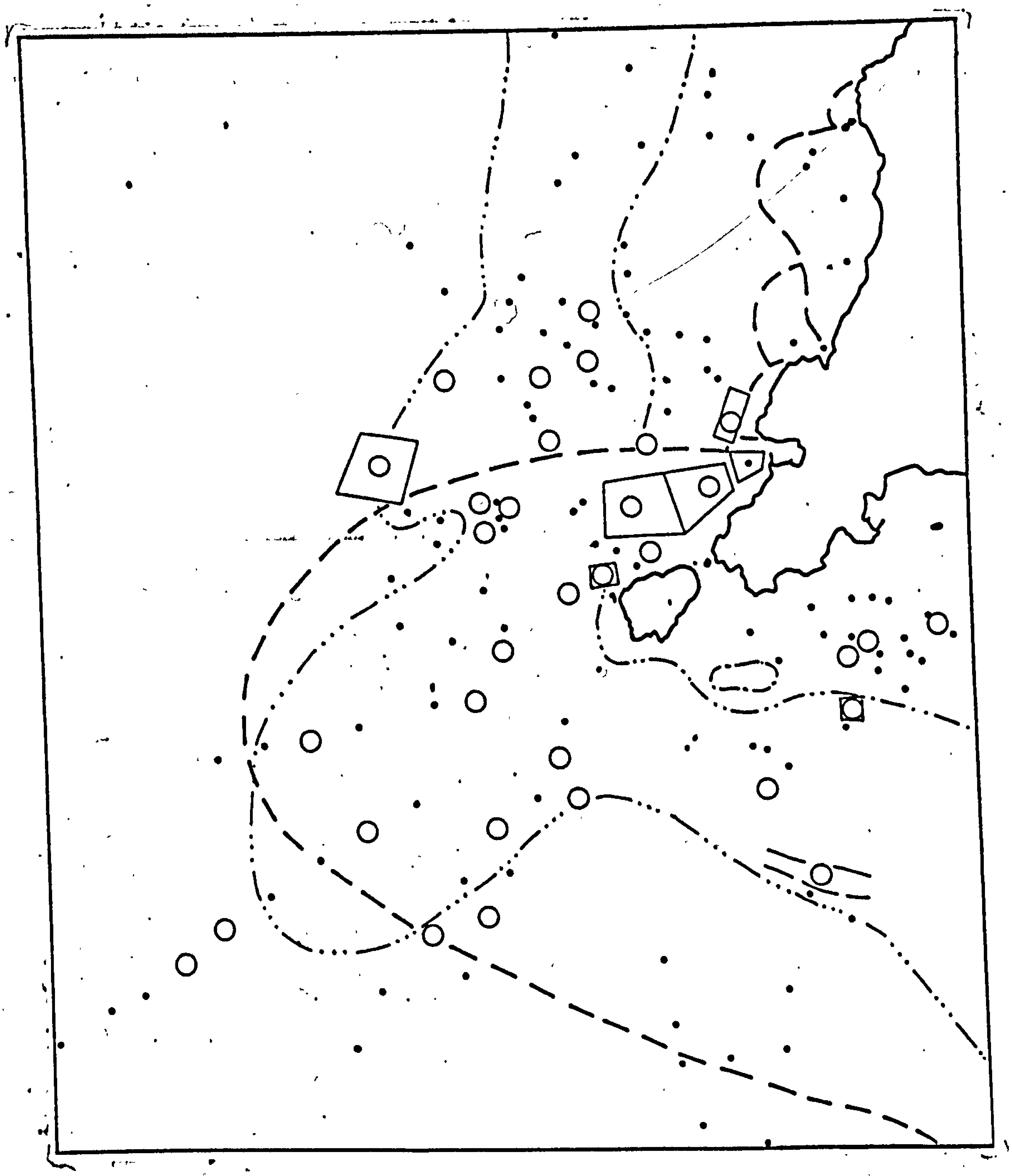
Map. 29. Scrupocellaria reptans. Offshore distribution within the area sampled.
O = locality at which S.reptans was obtained,
• = locality at which S.reptans was not obtained, rest as Map 1.



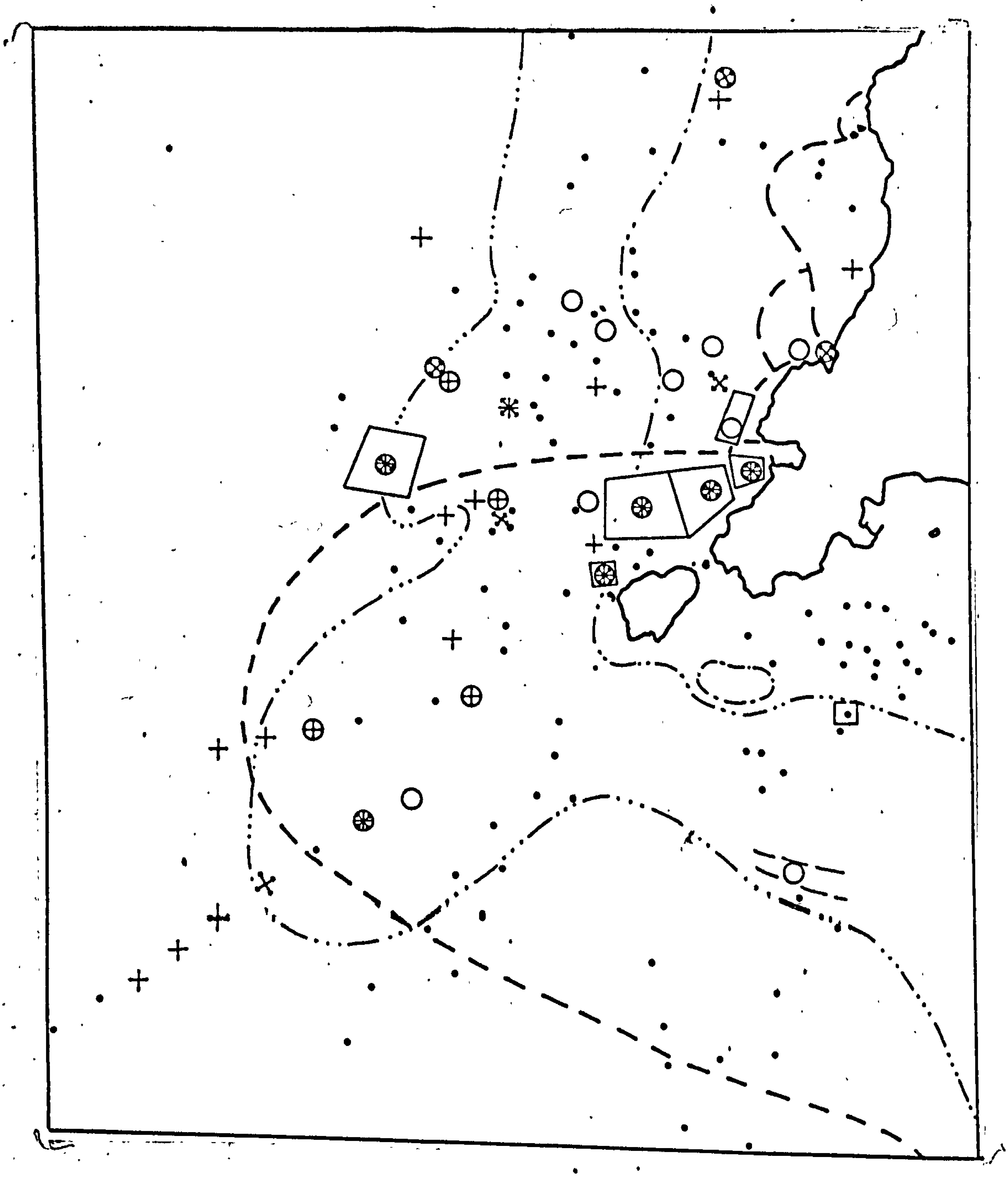
Map 30. Beania mirabiles. Offshore distribution within the area sampled.

O = locality at which B.mirabiles was obtained,

• = locality at which B.mirabiles was not obtained, rest as Map 1.



Map. 31. Bicellariella ciliata. Distribution within the area sampled.
o= locality at which B.ciliata was obtained,
• = locality at which B.ciliata was not obtained, rest as
Map.1.



Map. 32. Bugula avicularia, ,Bugula plumosa, Bugula flabellata. Offshore distribution within the area sampled.

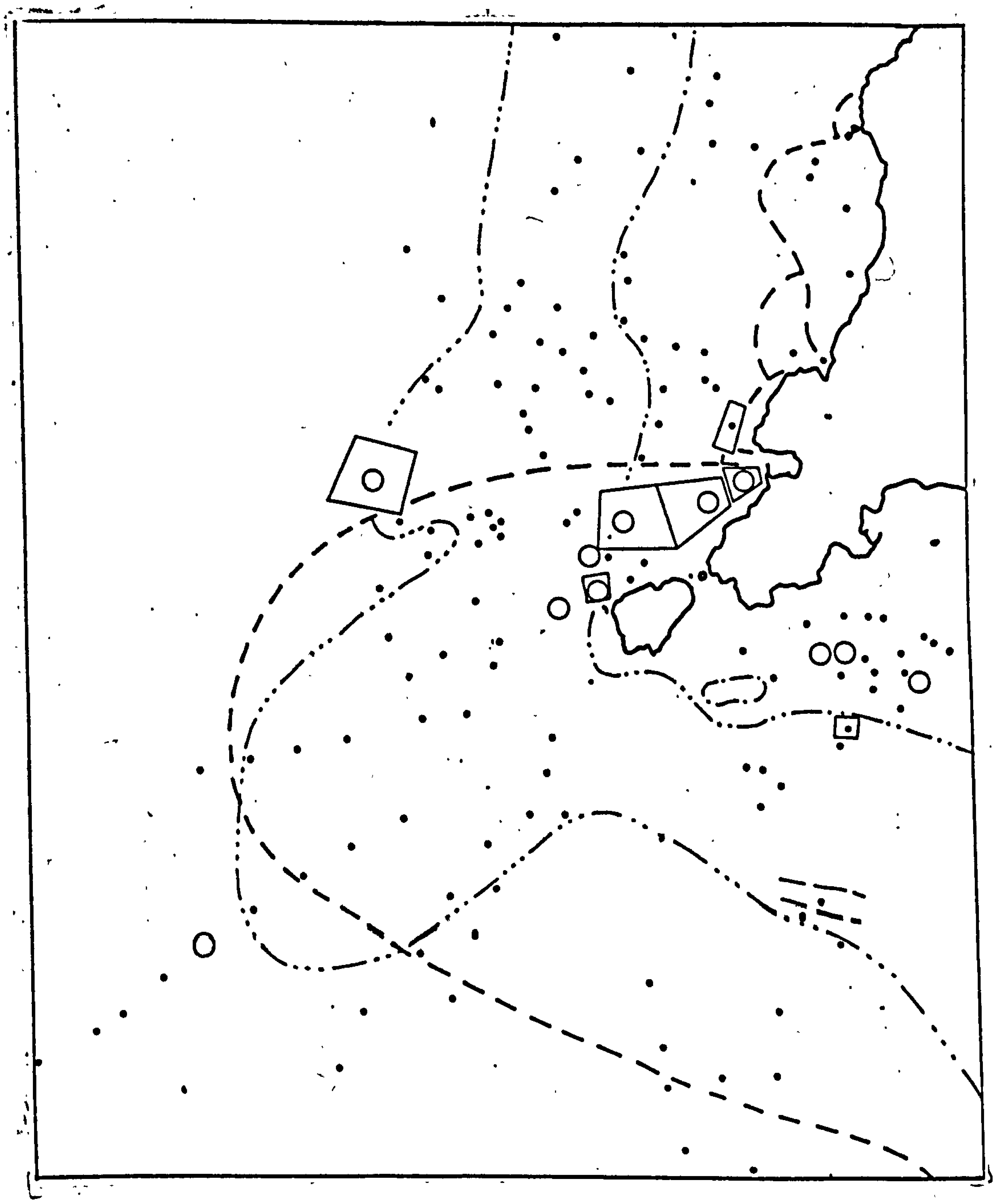
+ = locality at which B.avicularia was obtained,.

x = locality at which B.plumosa was obtained,

o = llocality at which B.flabellata was obtained,

• = locality at which none of these species was obtained,

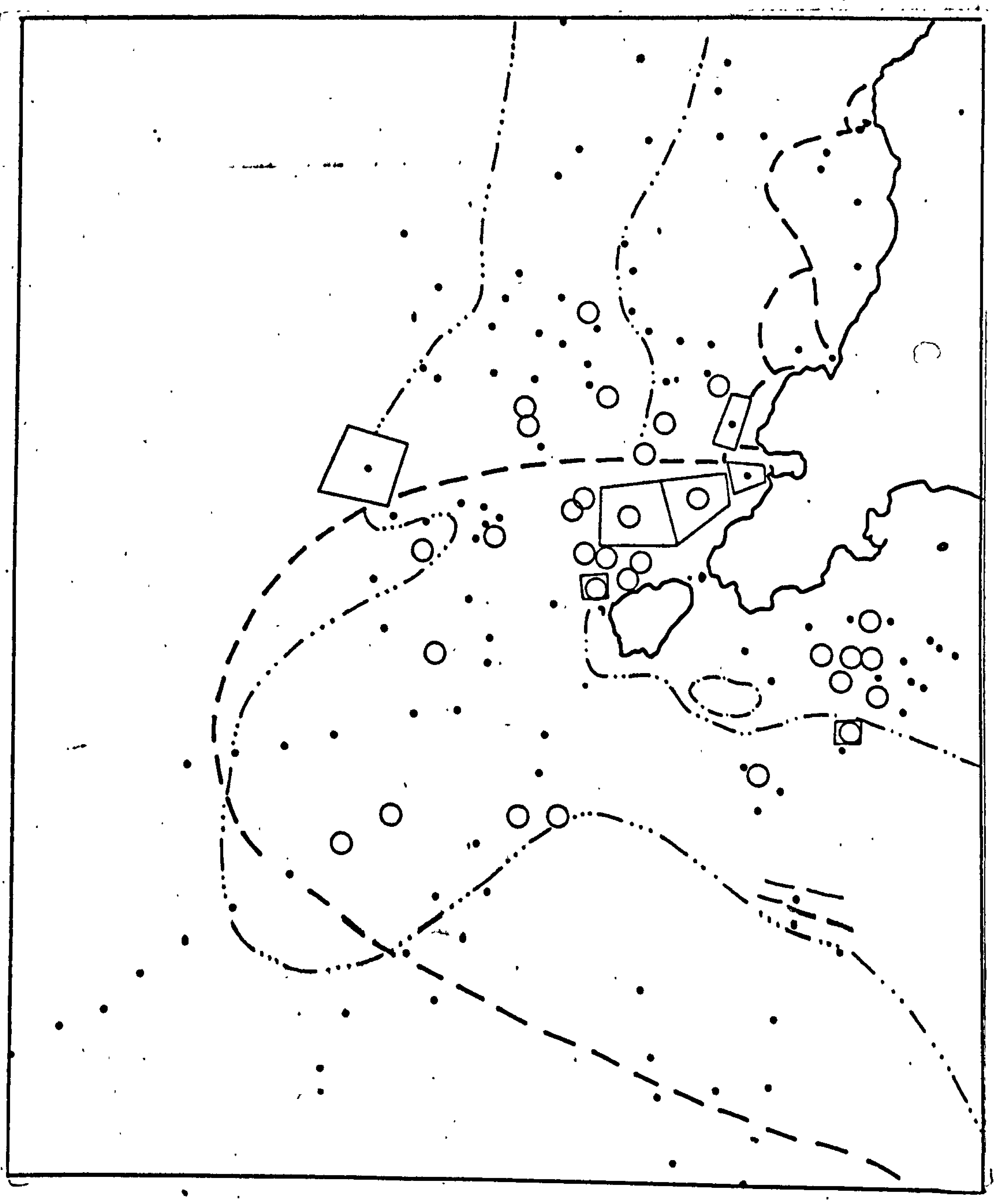
rest as Map 1.



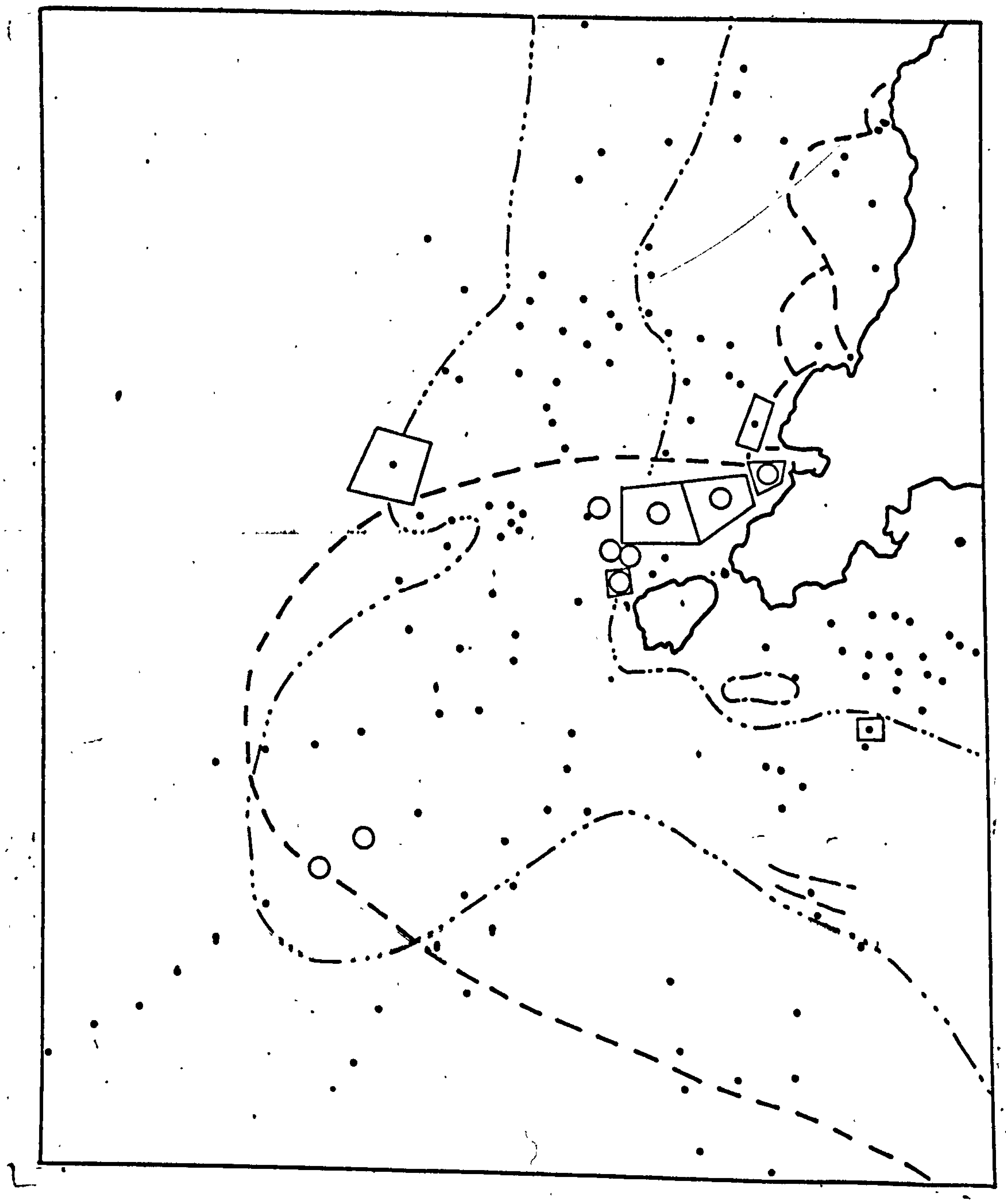
Map. 33. Membraniporella nitida. Offshore distribution within the area sampled.

o = locality at which M.nitida was obtained,

• = locality at which M.nitida was not obtained, rest as Map 1.



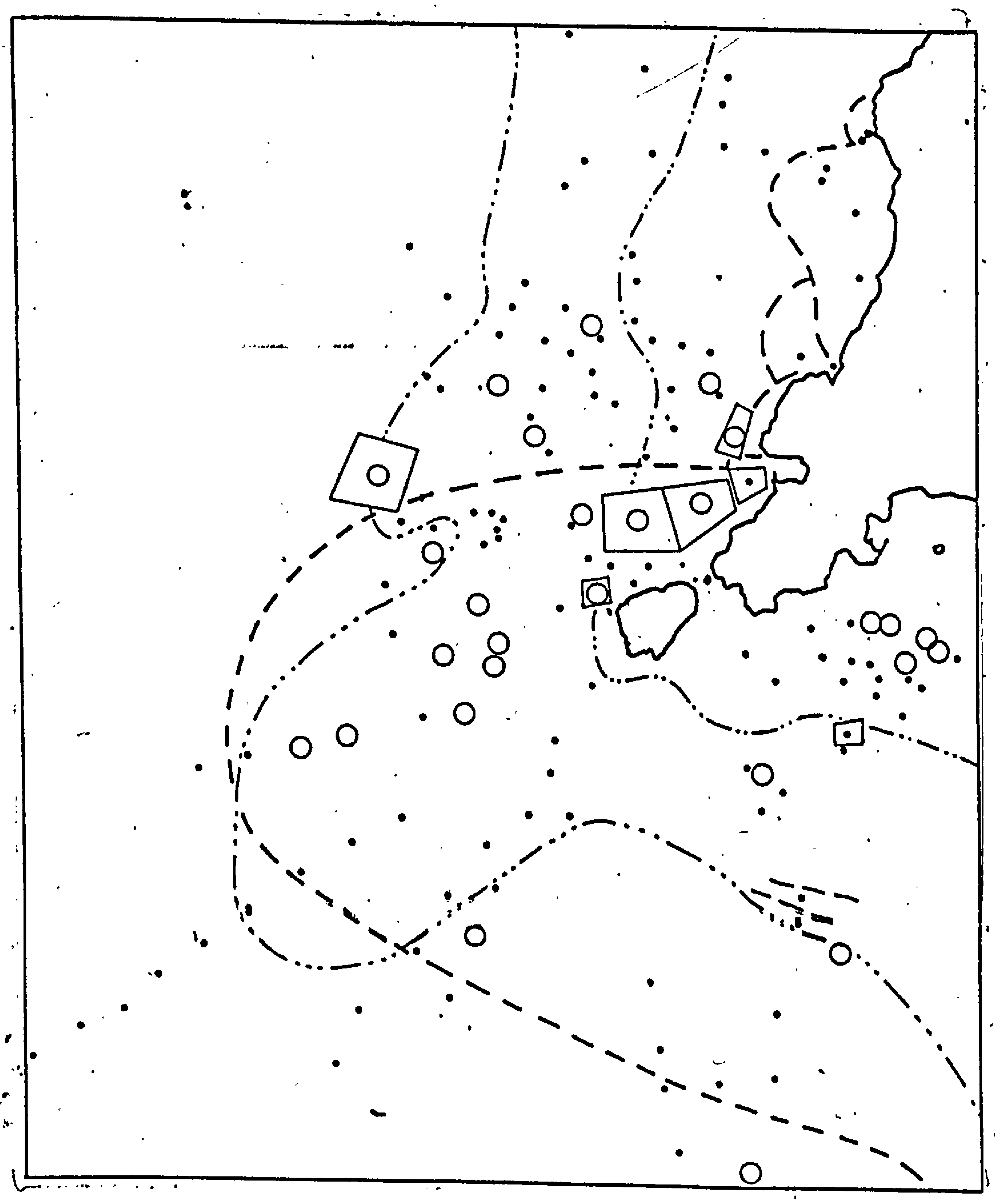
Map. 34. Cribrilaria radiata. Offshore distribution within the area sampled.
o = locality at which C. radiata was obtained,
• = locality at which C. radiata was not obtained, rest as Map 1.



Map 35. Figularia figularis. Offshore distribution within the area sampled.

○ = locality at which F.figularis was obtained,

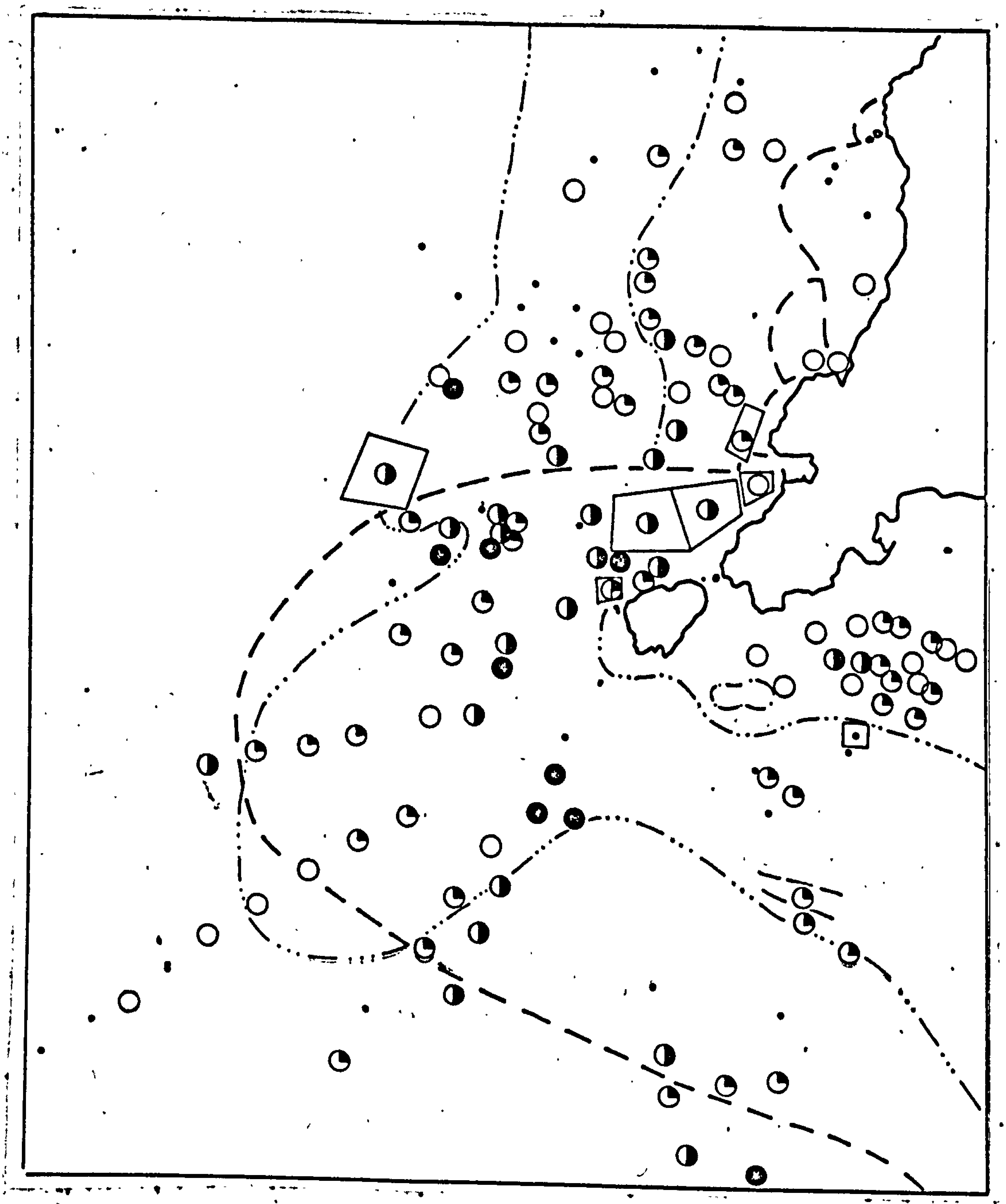
• = locality at which F.figularis was not obtained, rest as Map 1.



Map. 36. Hippothoa divaricata. Offshore distribution in the area sampled.

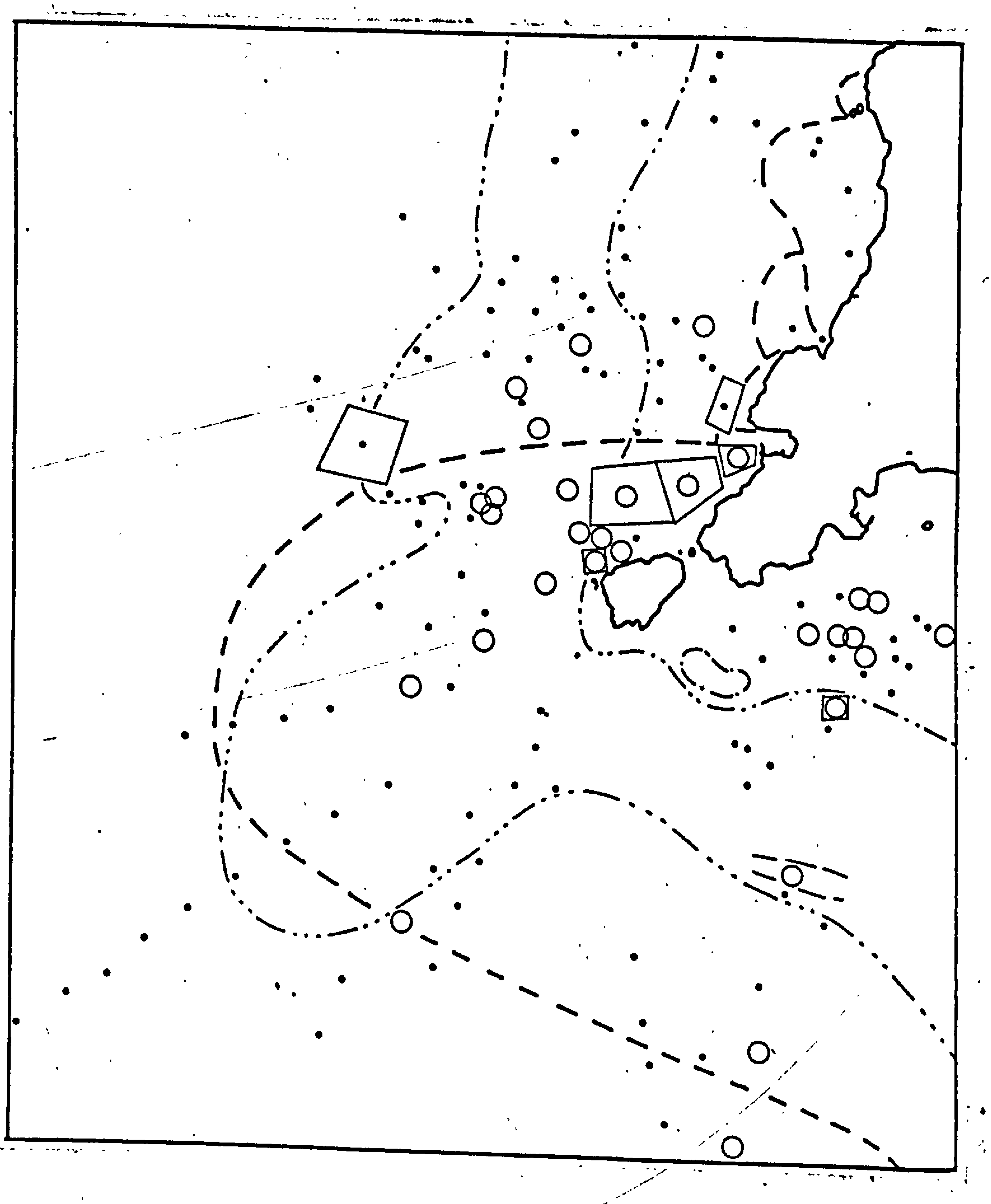
o = locality at which H. divaricata was obtained,

• = locality at which H. divaricata was not obtained, rest as Map. 1.



Map. 37.Hippothoa distans. Distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excuding erect zoophytes) suitable for ectoproct colonization.

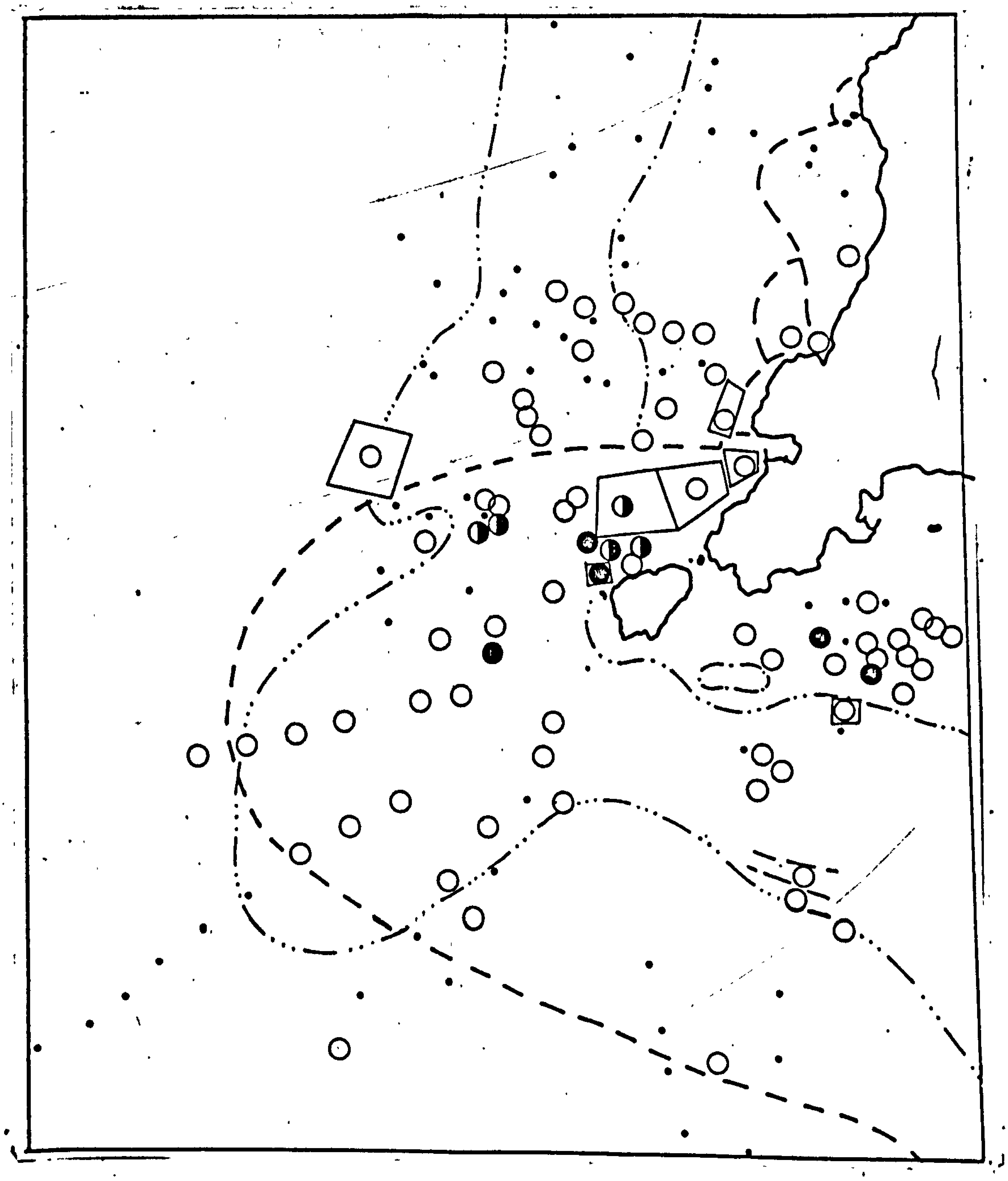
• = locality at which H.distans was not obtained,
○ = less than 1 colony per 1000cm^2,
◔ = 1 - 5 colonies per 1000cm^2,
◑ = 5 - 10 colonies per 1000cm^2,
● = more than 10 colonies per 1000cm^2, rest as Map 1.



Map. 38. Hippothoa hyalina. Offshore distribution within the area sampled.

O = locality at which H.hyalina was obtained,

• = locality at which H.hyalina was not obtained, rest as Map 1.



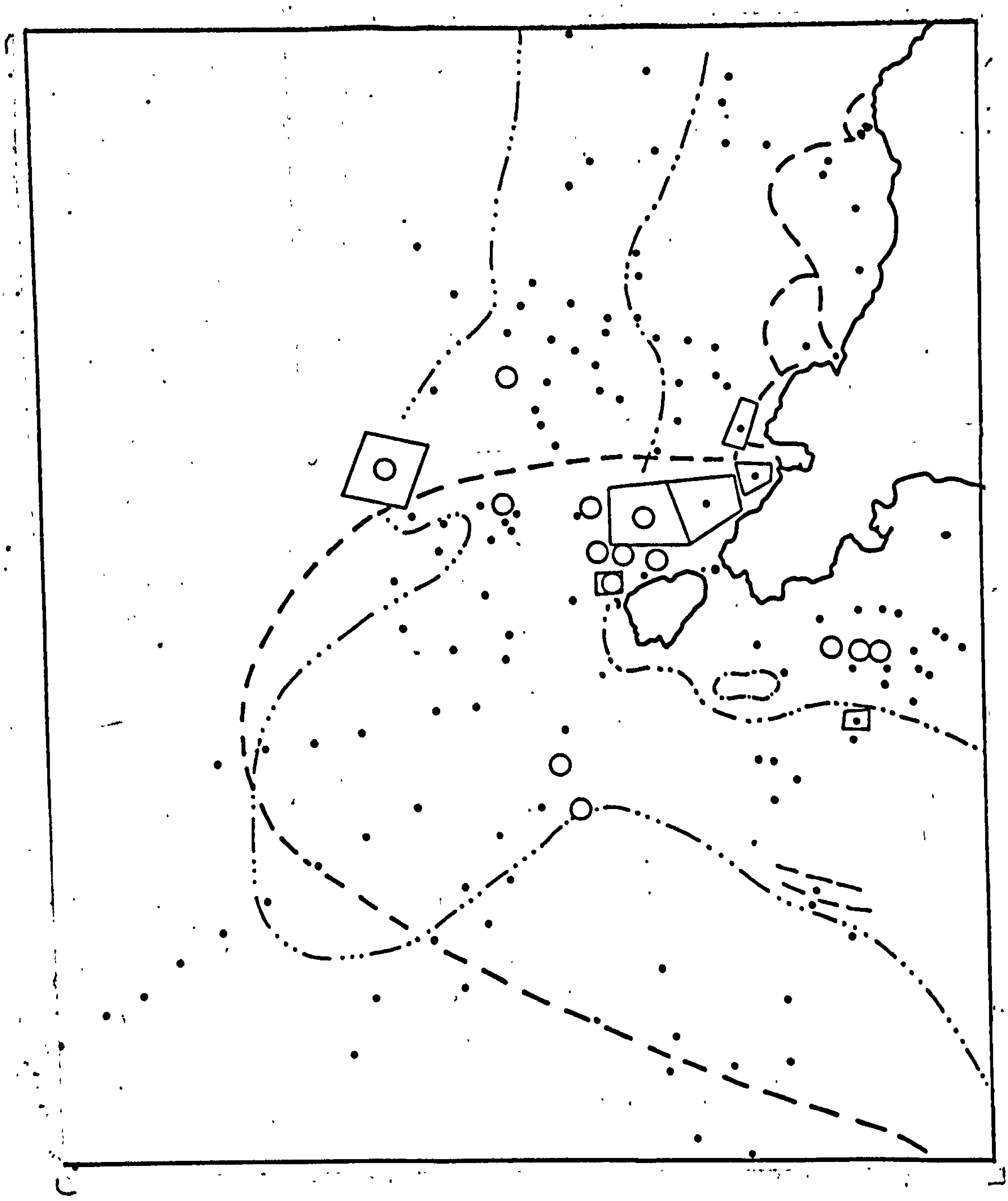
Map. 39. Chorizopora brongniarti. Offshore distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

• = locality at which ~~C. brongniarti~~ was not obtained,

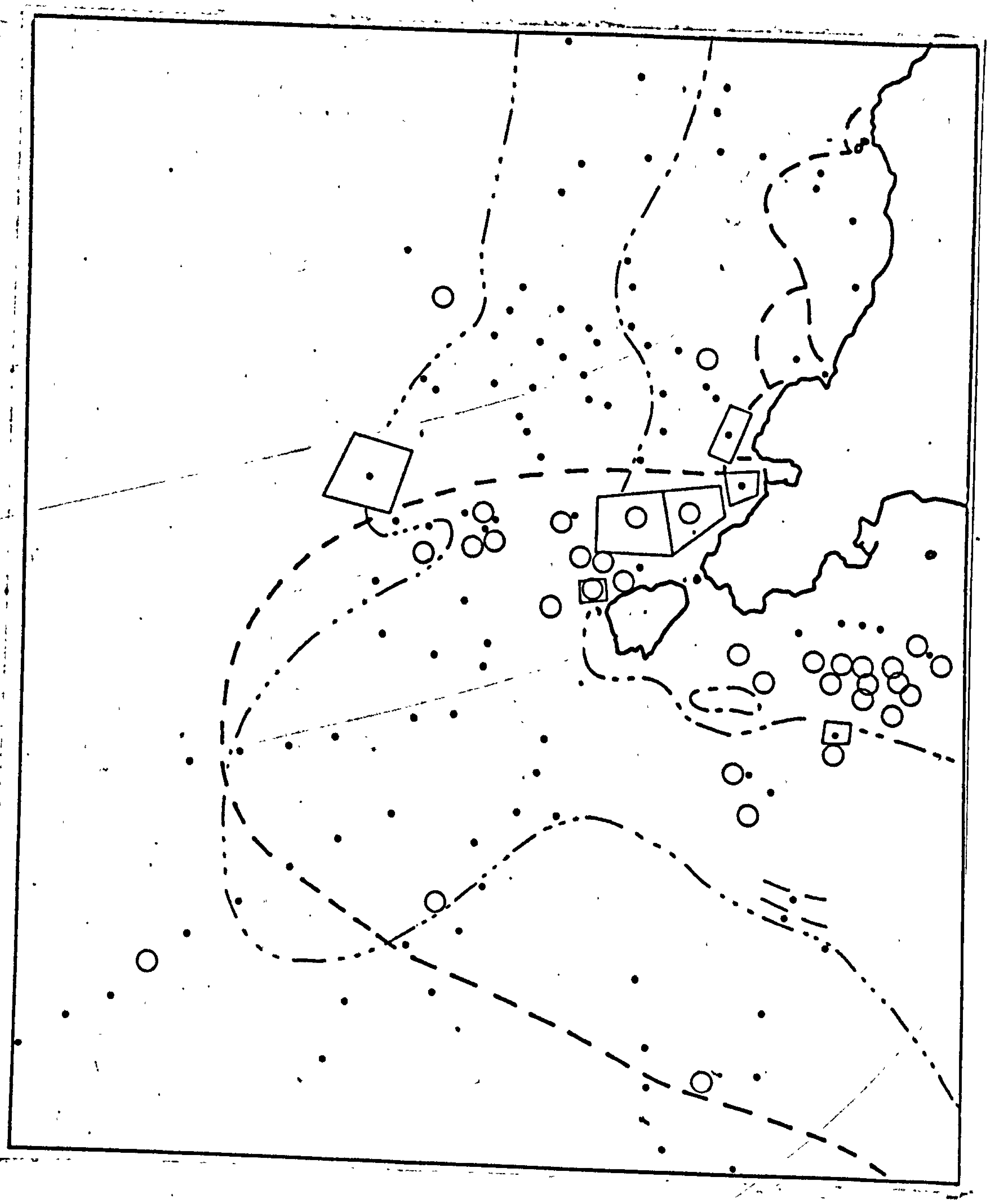
O = less than 5 colonies per 1000cm^2,

◑ = 5 - 10 colonies per 1000cm^2,

● = more than 10 colonies per 1000cm^2, rest as Map 1.



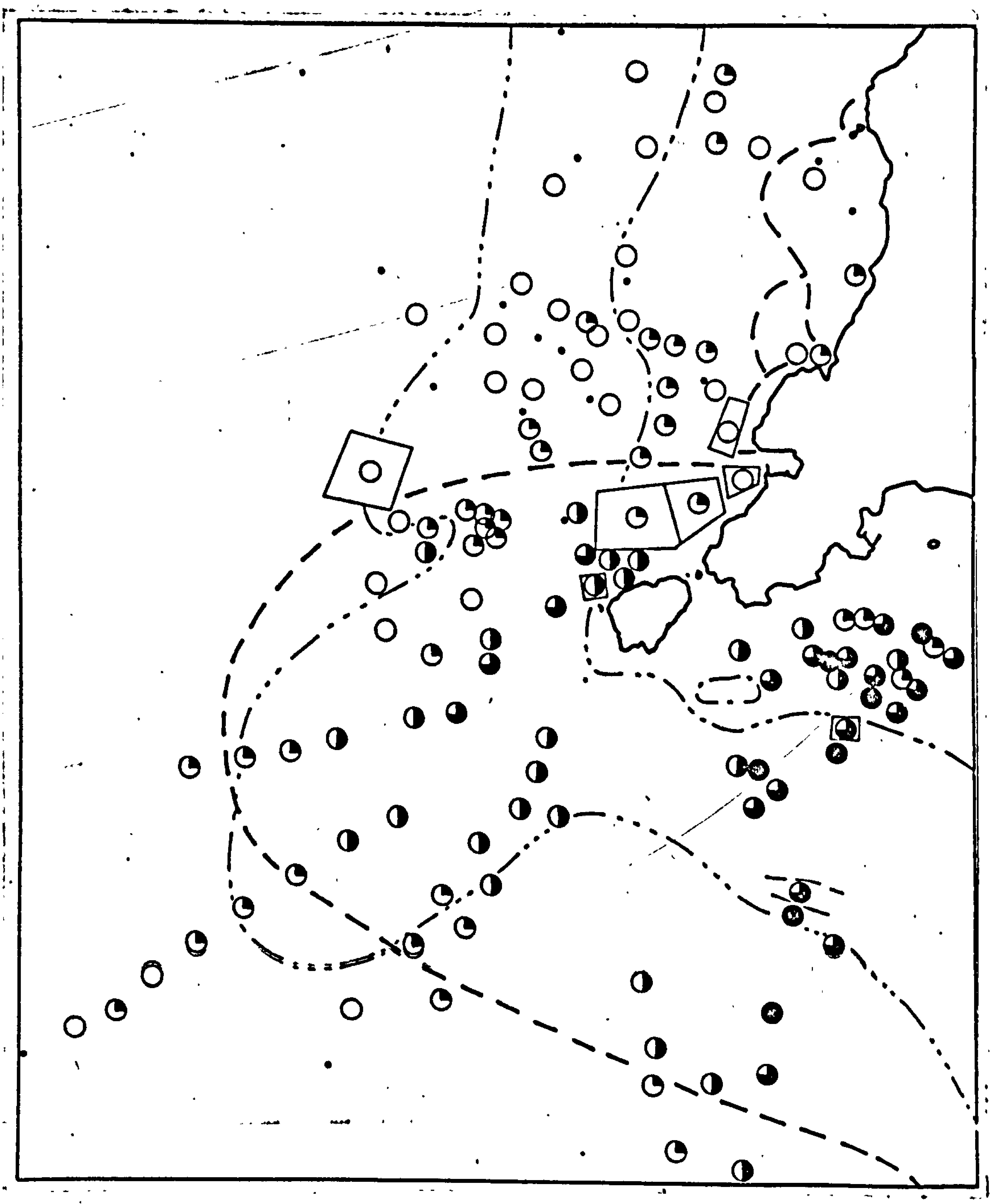
Map 40. Schizotheca fissa. Distribution within the area sampled.
• = locality at which S.fissa was not obtained,
O = locality at which S.fissa was obtained, rest as Map 1.



Map. 41. <u>Reptadeonella</u> <u>violacea</u>. Distribution within the area sampled.

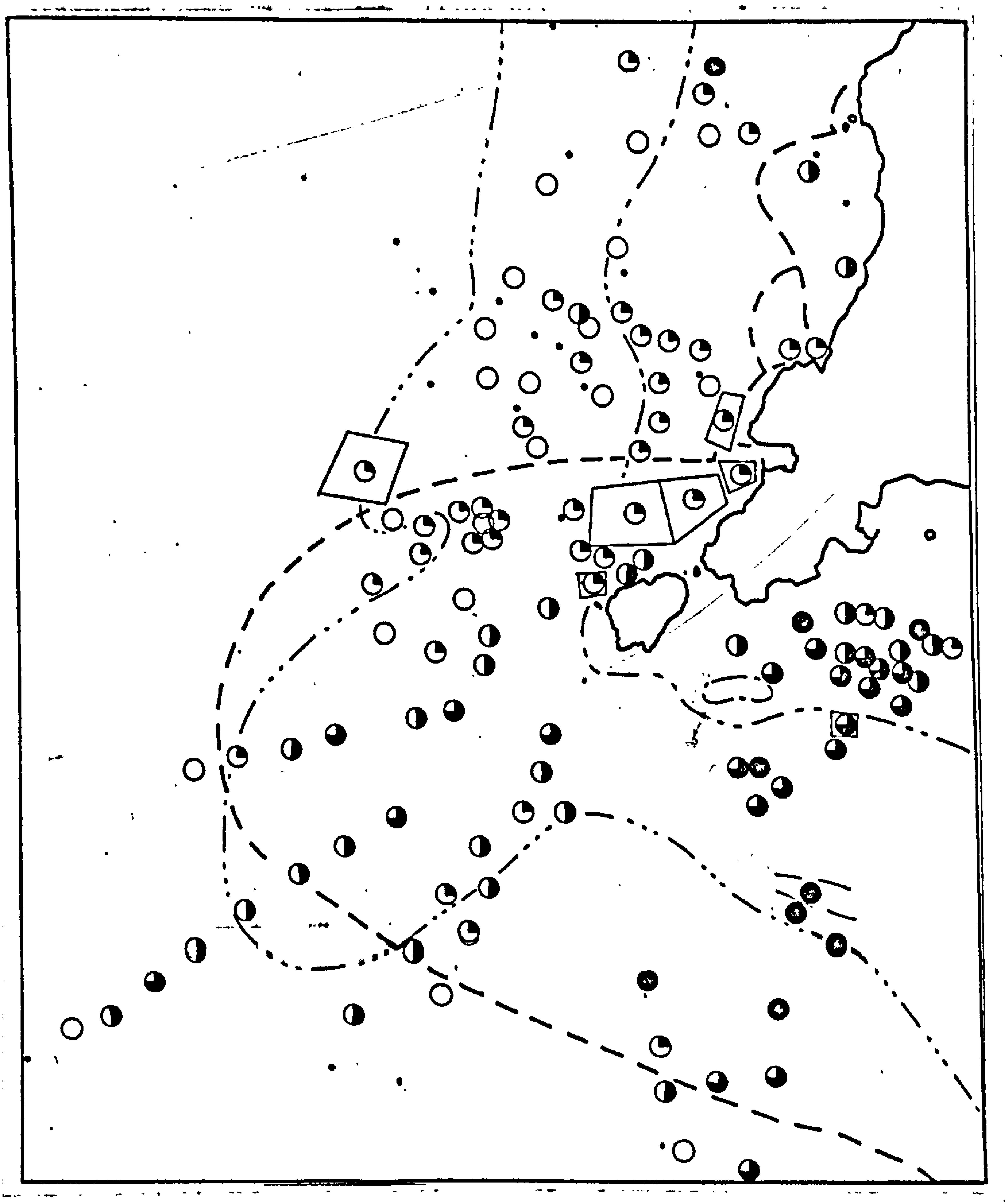
• = locality at which <u>R.violacea</u> was not obtained,

O = locality at which <u>R.violacea</u> was obtained, rest as Map 1.



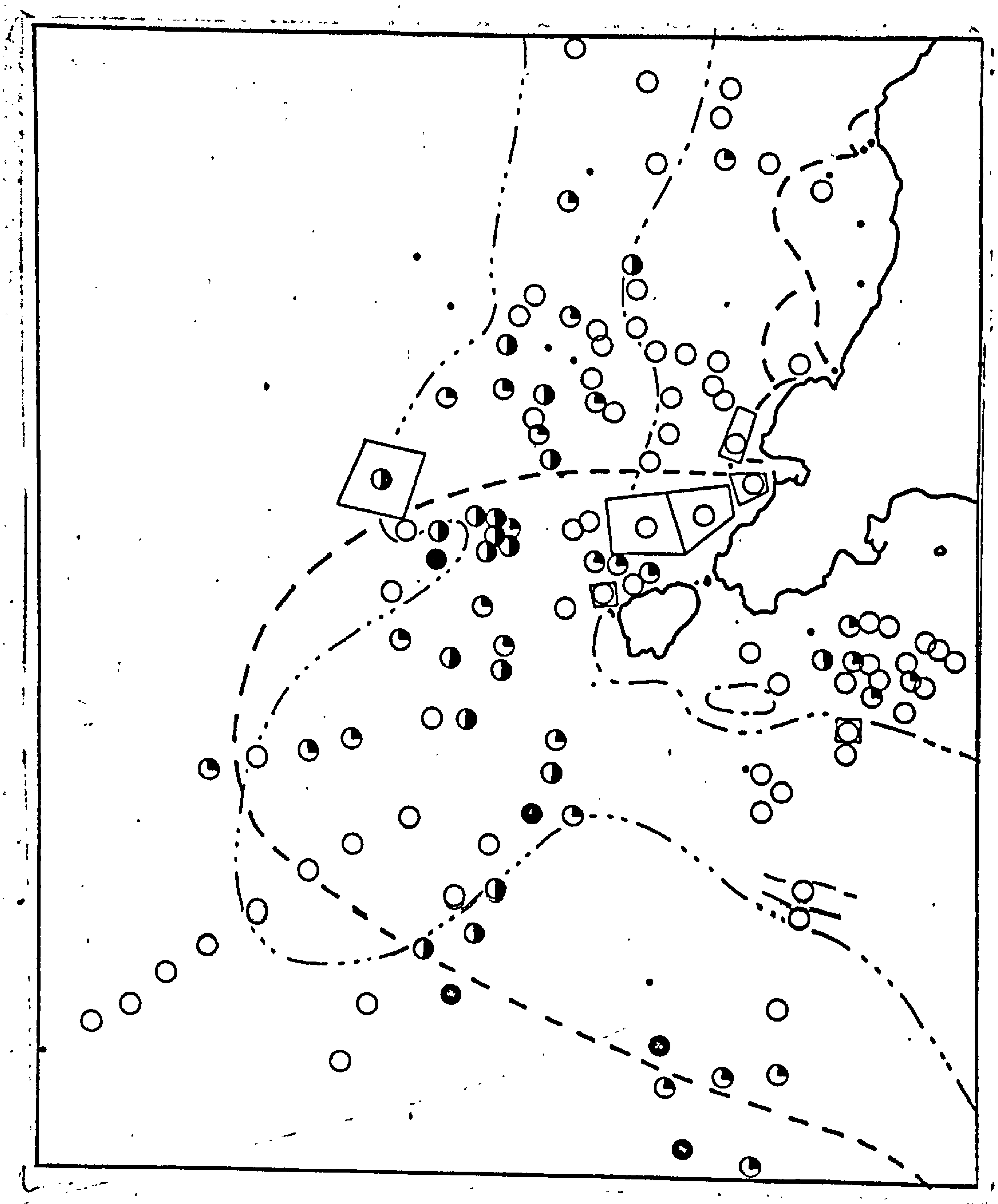
Map. 42. Escharella immersa. Offshore distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2, surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

- • = locality at which E.immersa was not obtained,
- ○ = less than 5 colonies per 1000 cm^2,
- ◔ = 5 -20 colonies per 1000 cm^2,
- ◑ = 20 -50 colonies per 1000 cm^2,
- ◕ = 50 - 100 colonies per 1000 cm^2,
- ● = more than 100 colonies per 1000 cm^2, rest as Map 1.



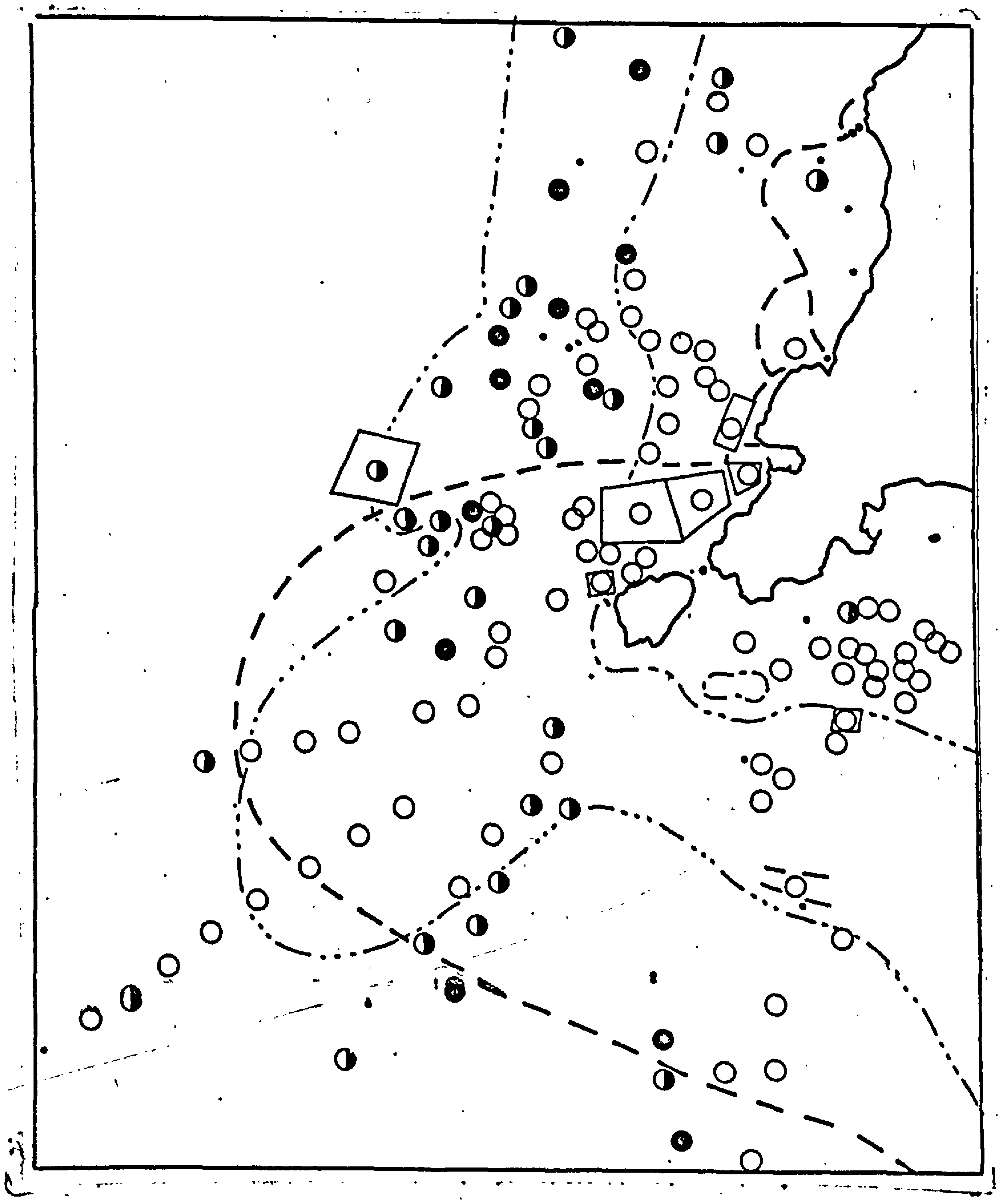
Map. 43. Escharella immersa. Relative importance in the ectoproct population within the area sampled.

• = locality at which E.immersa was not obtained,
○ = locality at which less than 5 %,
◔ = locality at which from 5 - 10 %,
◑ = locality at which from 10 - 20 %,
◕ = locality at which from 20 - 40 %,
● = locality at which more than 40 % of the ectoprocts are E.immersa, rest as Map 1.



Map. 44. Escharella ventricosa. Offshore distribution and abundance within the area sampled in terms of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

• = locality at which E.ventricosa was not obtained,
○ = less than 5 colonies per 1000cm^2,
◔ = 5 - 10 colonies per 1000 cm^2,
◑ = 10 - 20 colonies per 1000 cm^2,
● = more than 20 colonies per 1000 cm , rest as Map 1.



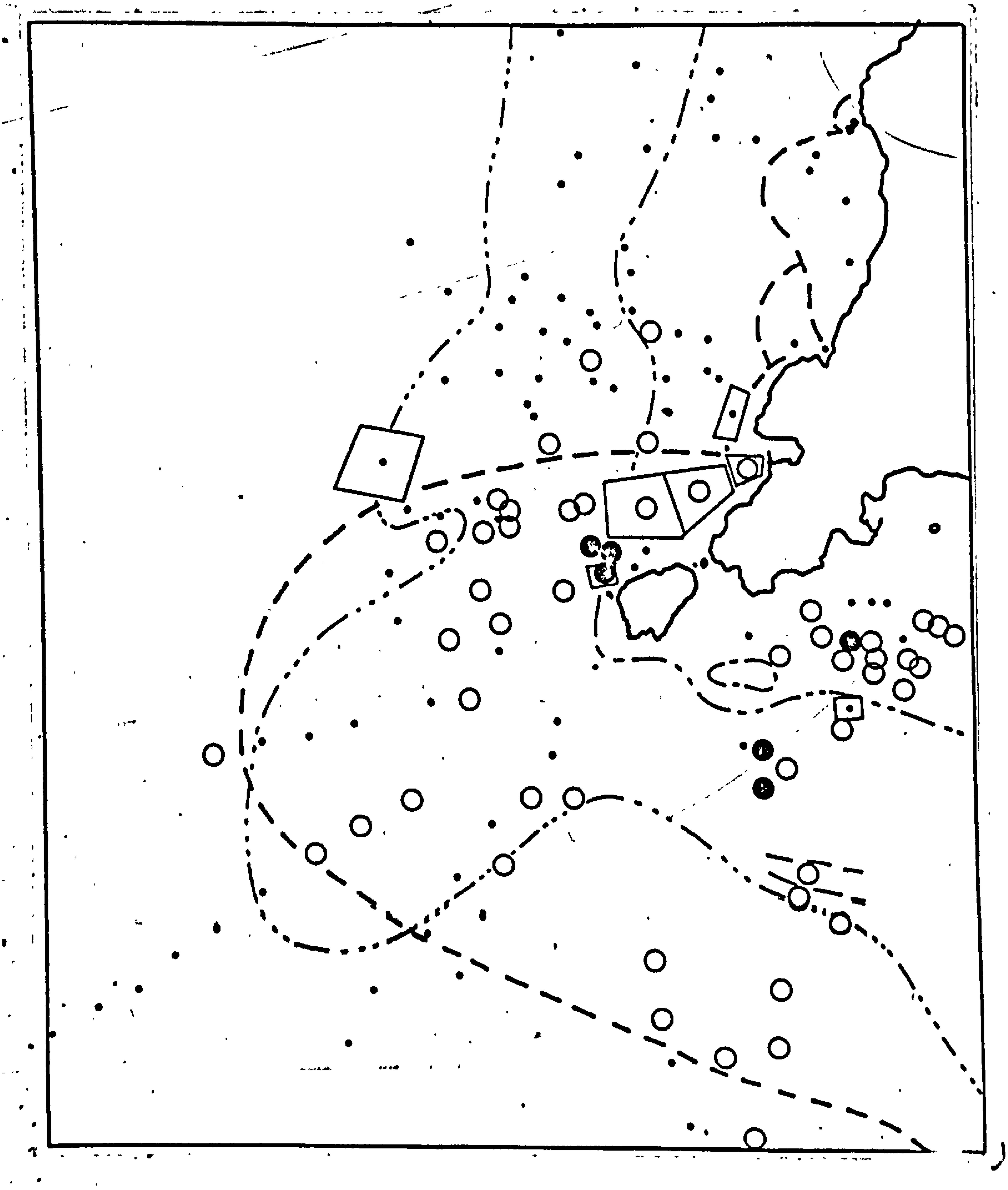
Map. 45. Escharella ventricosa. Relative impoBtance in the ectoproct population within the area sampled.

• = locality at which E.ventricosa was not obtained

O = locality at which less than 5 %,

◐ = locality at which from 5 - 10 %,

● = locality at which more than 10 % of the ectoprocts are E.ventricosa, rest as Map 1.

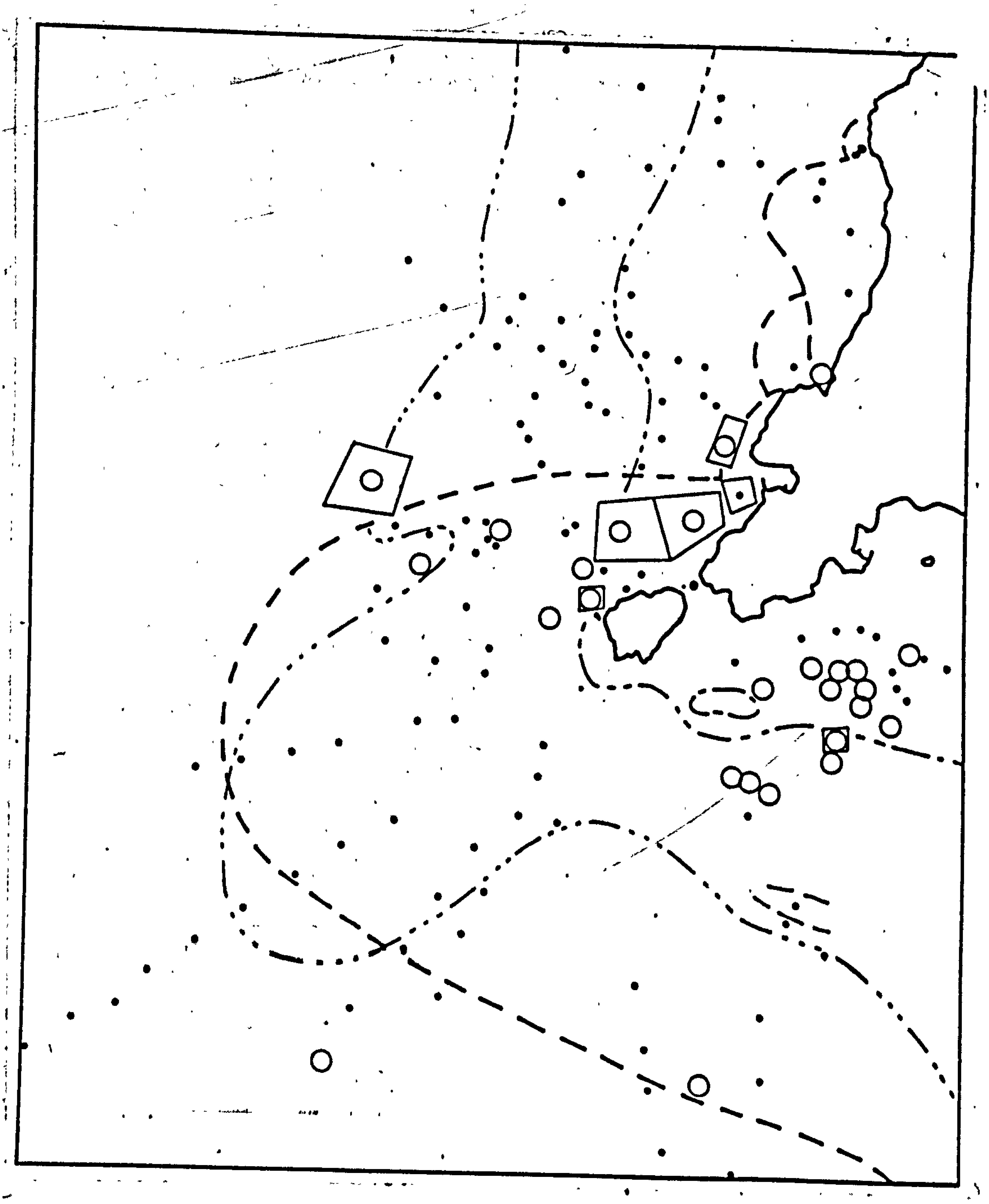


Map 46. Escharella variolosa. 'Offshore distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

• = locality at which E. variolosa was not obtained,

○ = less than 5 colonies per 1000 cm^2,

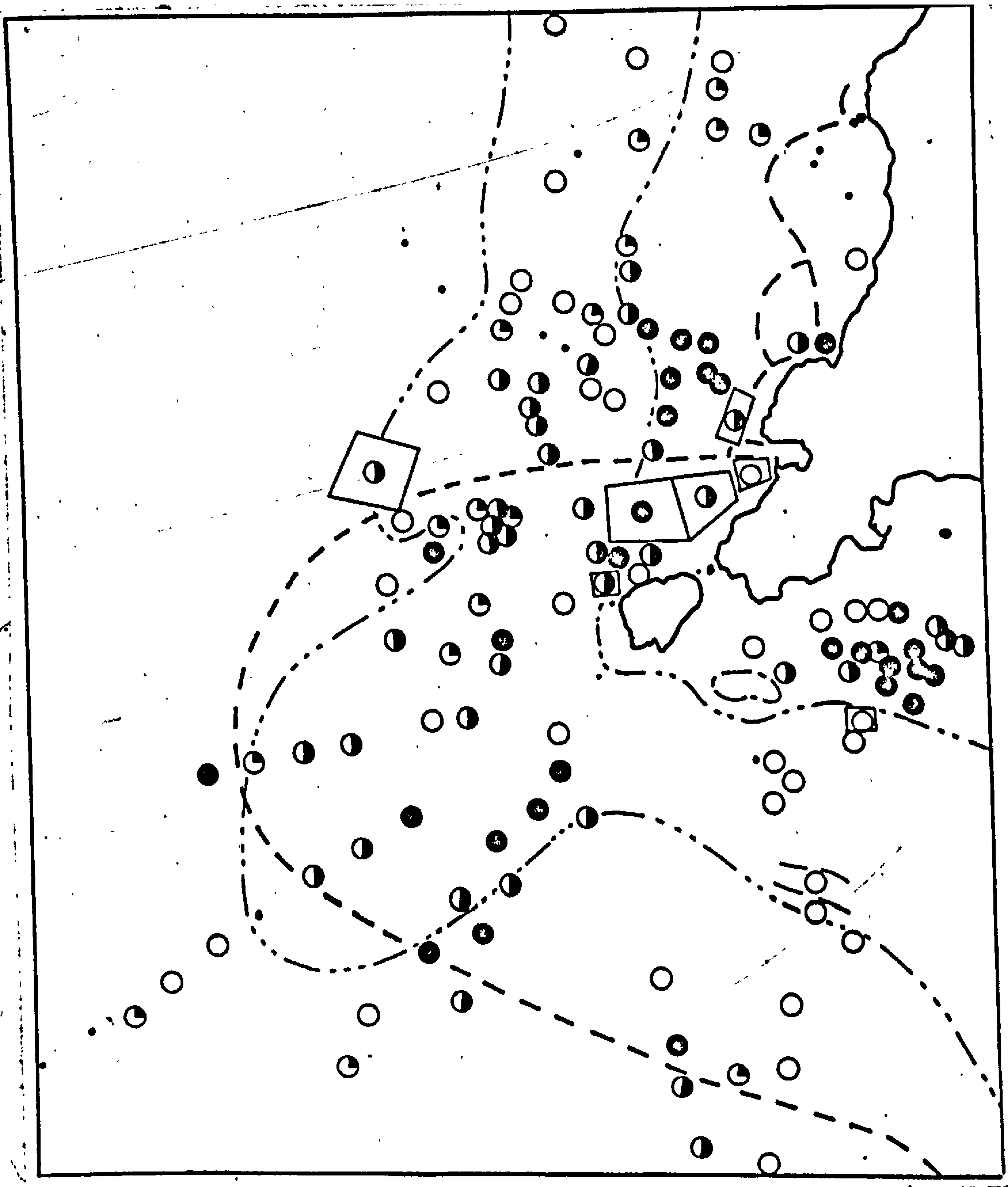
● = more than 5 colonies per 1000 cm^2, rest as Map 1.



Map 47. Schizoporella unicornis. Offshore distribution within the area sampled.

• = locality at which S.unicornis was not obtained,

O = locality at which S.unicornis was obtained, rest as Map 1.



Map 48. Schizomavella auriculata. Distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

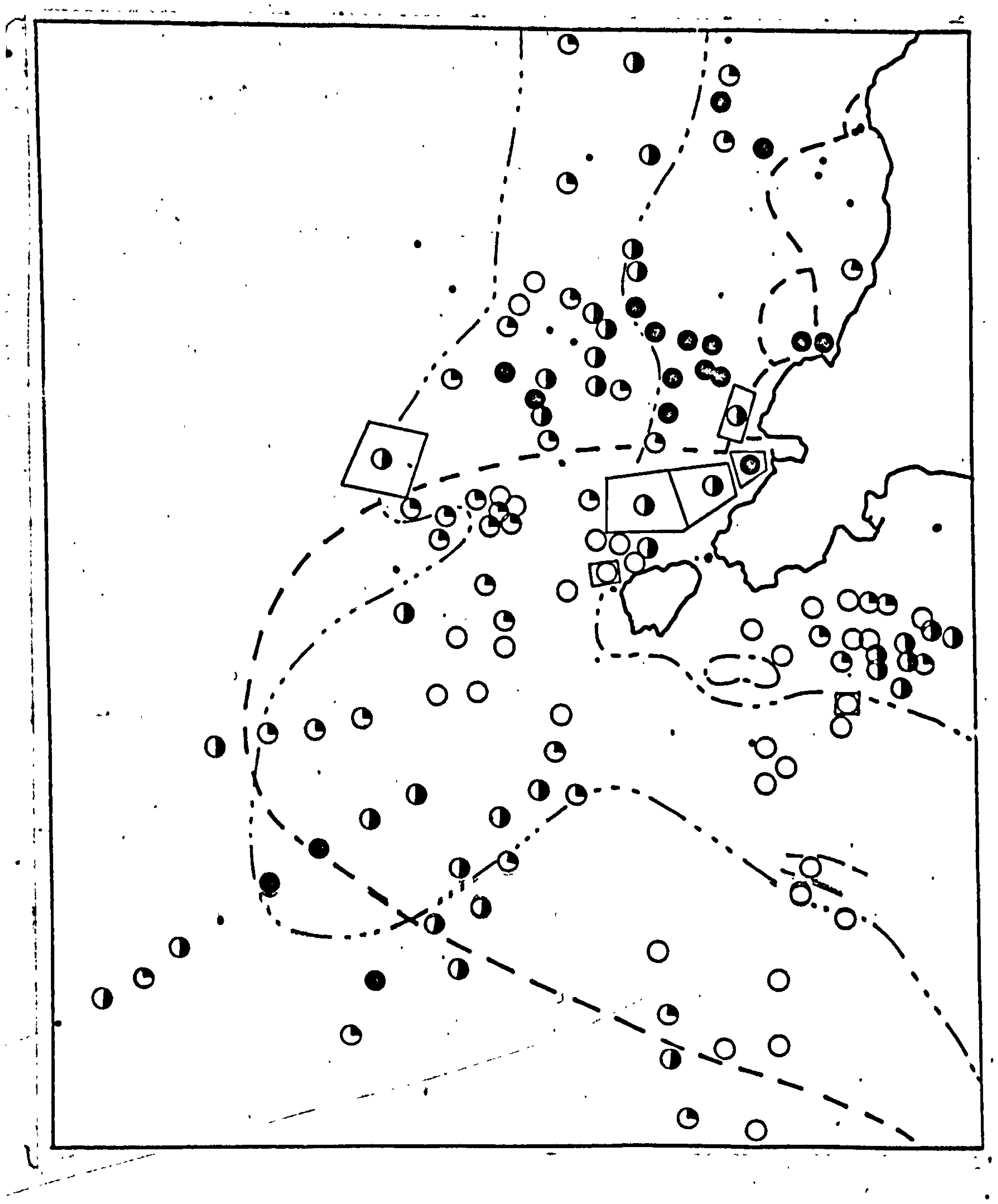
• = locality at which S. auriculata was not obtained,

○ = less than 5 colonies per 1000 cm^2,

◔ = 5 - 10 colonies per 1000 cm^2,

◑ = 10 - 20 colonies per 1000 cm^2,

● = more than 20 colonies per 1000 cm^2, rest as Map 1.



Map 49. Schizomavella auriculata. Relative importance in the ectoproct population within the area sampled.

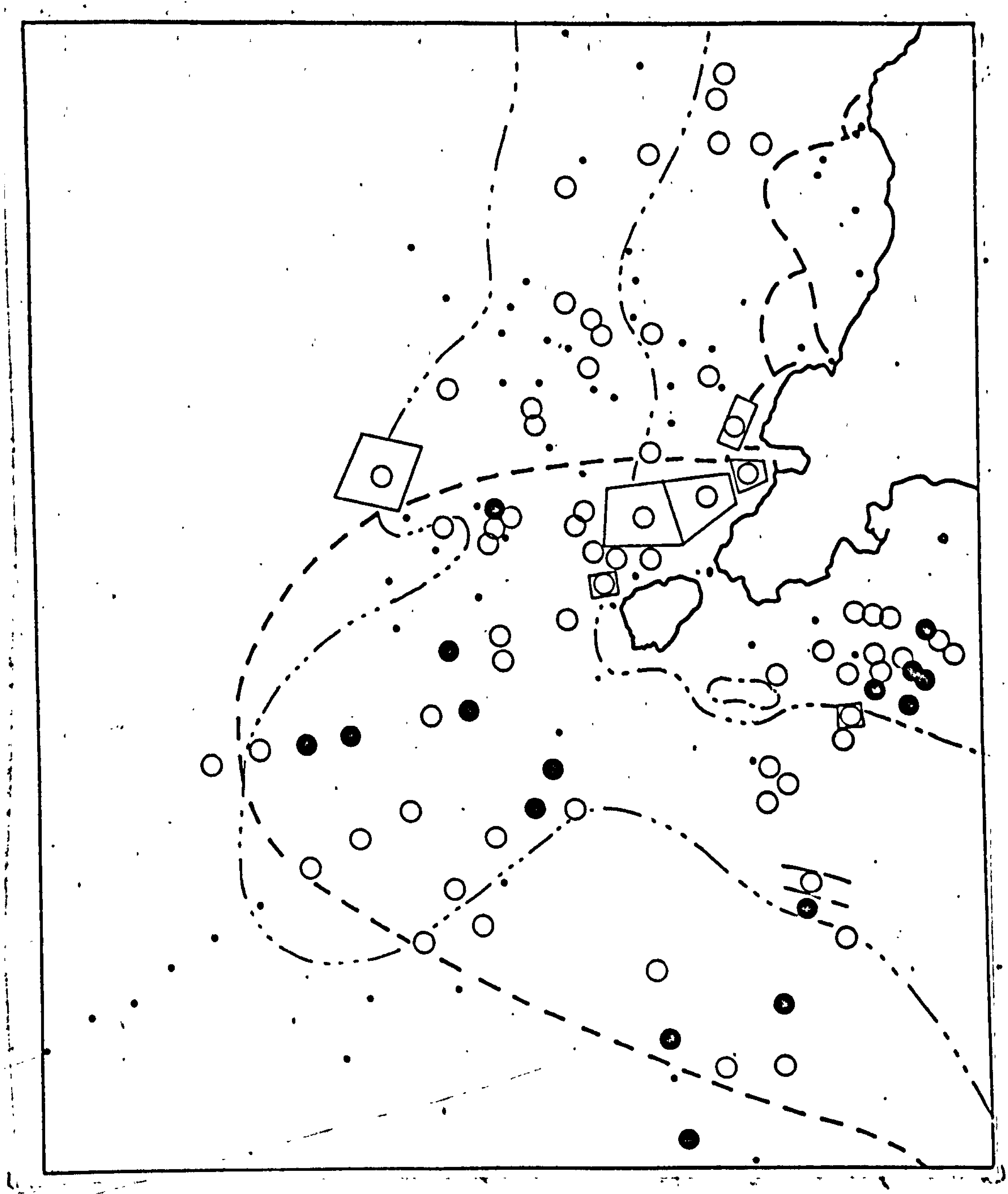
• = locality at which S. auriculata was not obtained,

○ = locality at which less than 5 %,

◔ = locality at which from 5 - 10 %,

◑ = locality at which from 10 - 20 %,

● = locality at which more than 20 % of the ectoprocts are S. auriculata, rest as Map 1.

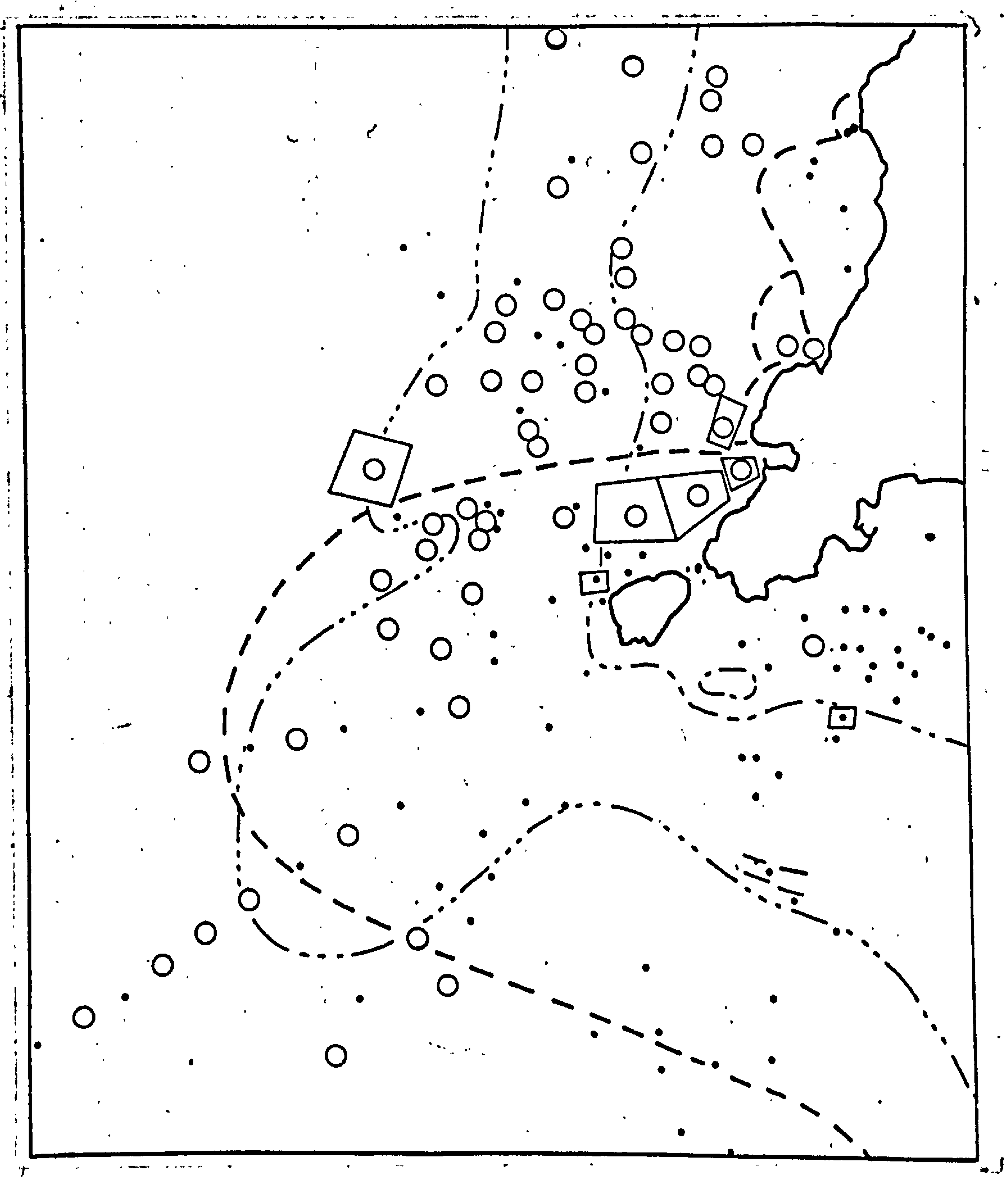


Map 50. Schizomavella linearis. Offshore distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

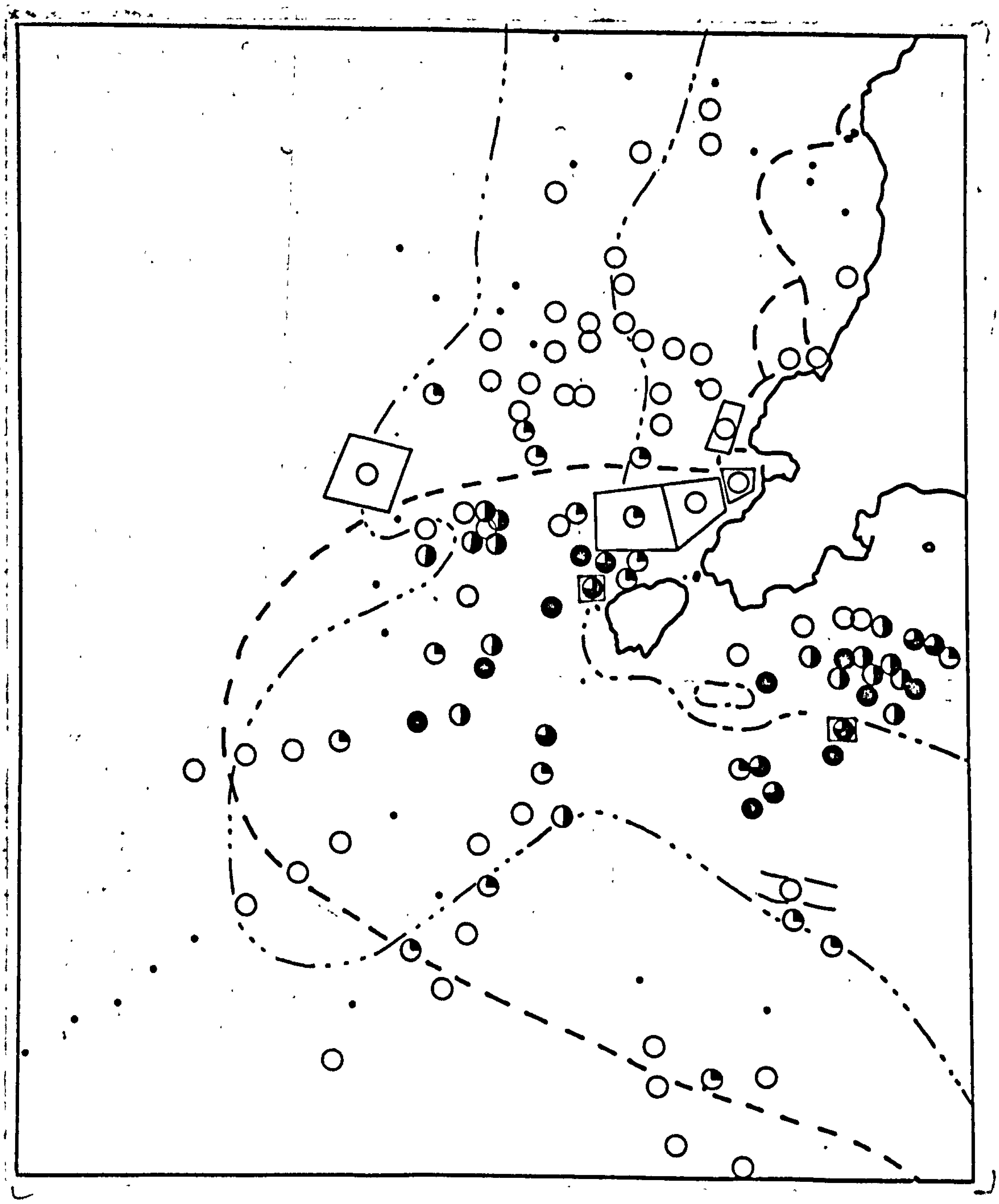
• = locality at which S. linearis was not obtained,

○ = less than 5 colonies per 1000^2 cm ,

● = more than 5 colonies per 1000^2 cm , rest as Map 1.

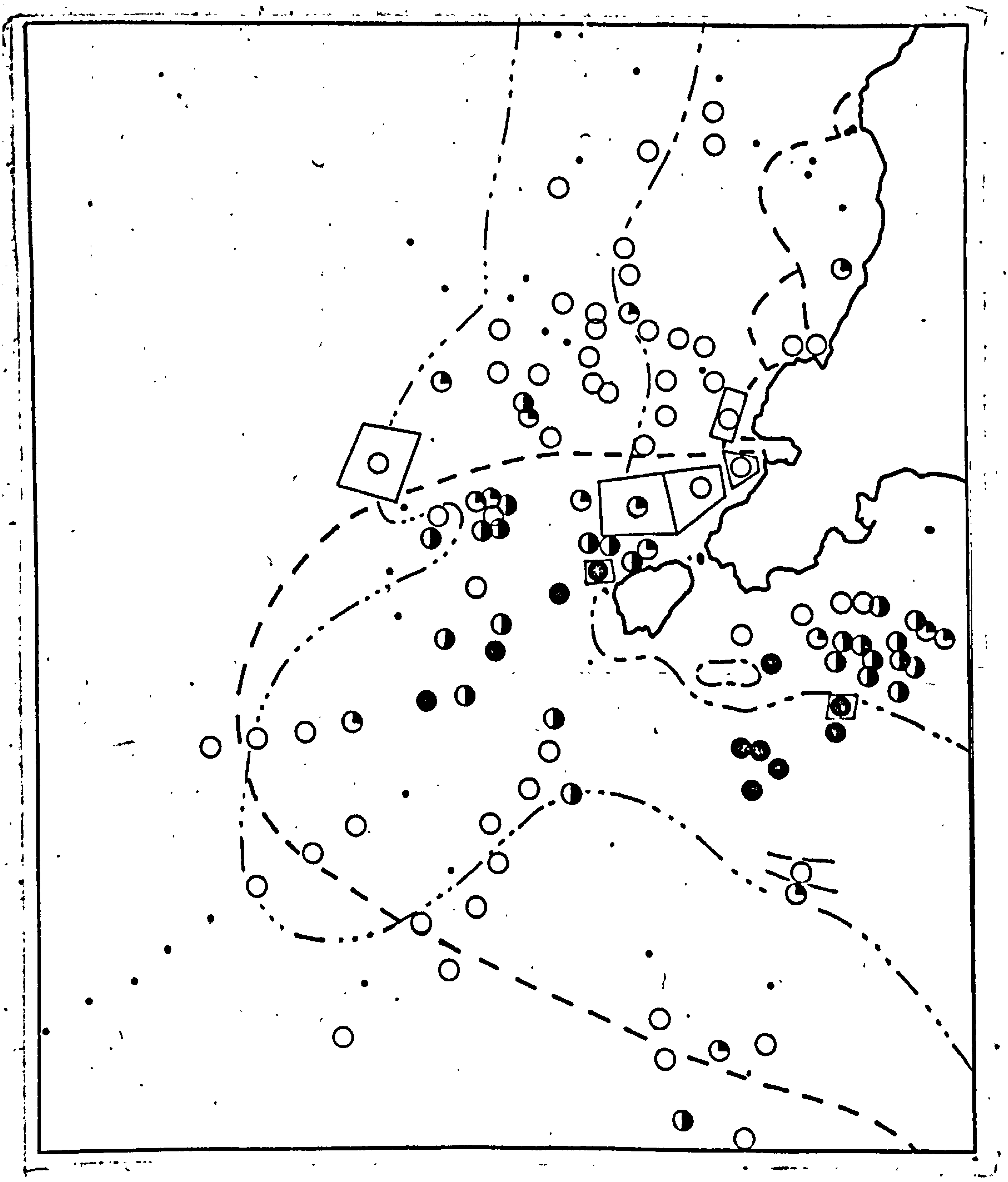


Map 51. Hipporina pertusa. Distribution within the area sampled.
• = locality at which H.pertusa was not obtained,
O = locality at which H.pertusa was obtained, rest as Map 1.



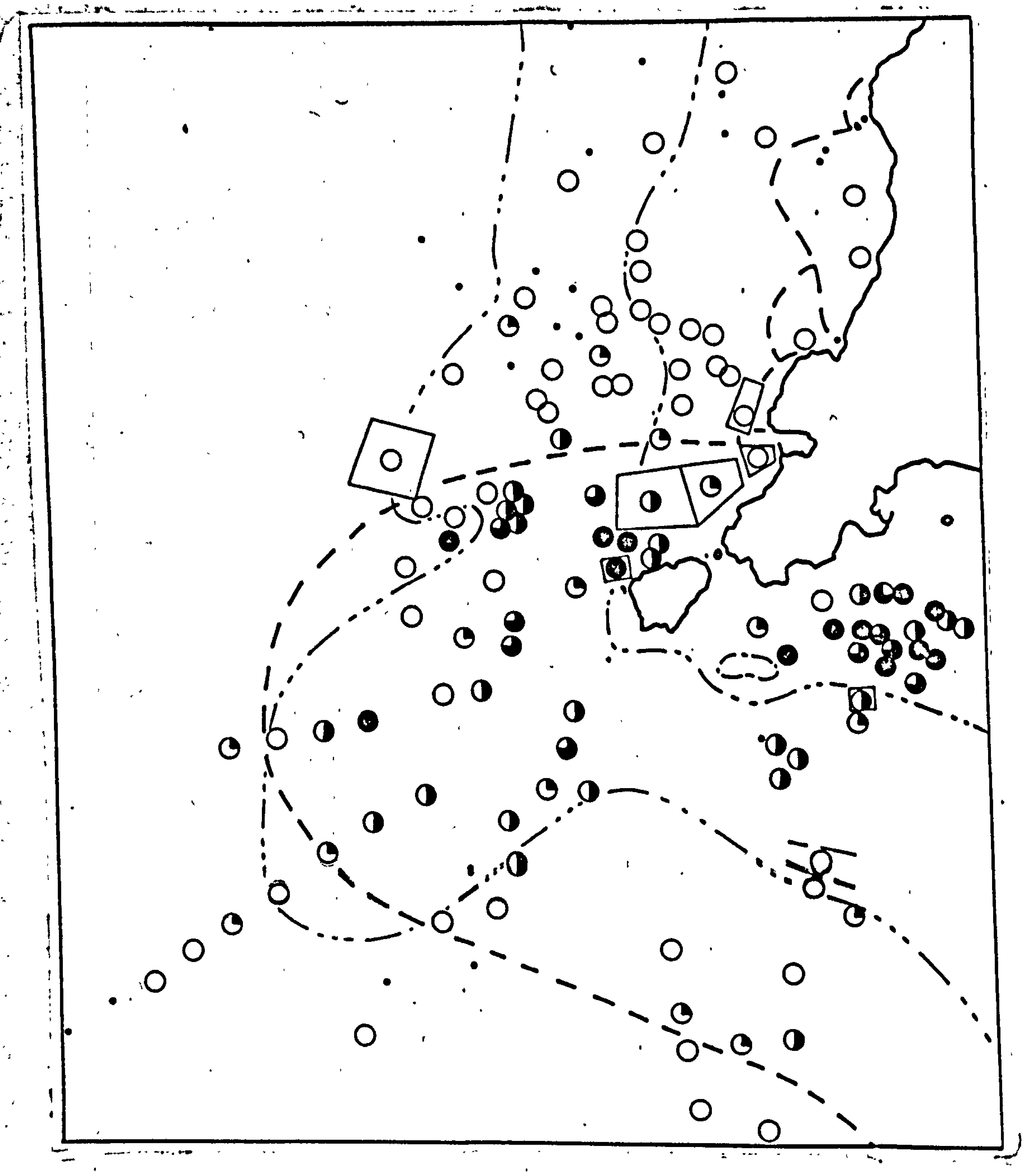
Map 52. Microporella ciliata. Offshore distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

- • = locality at which M.ciliata was not obtained,
- O = less than 5 colonies per 1000 cm^2,
- ◔ = 5 - 20 colonies per 1000 cm^2,
- ◑ = 20 - 50 colonies per 1000 cm^2,
- ◕ = 50 - 100 colonies per 1000 cm^2,
- ● = more than 100 colonies per 1000 cm^2, rest as Map 1.



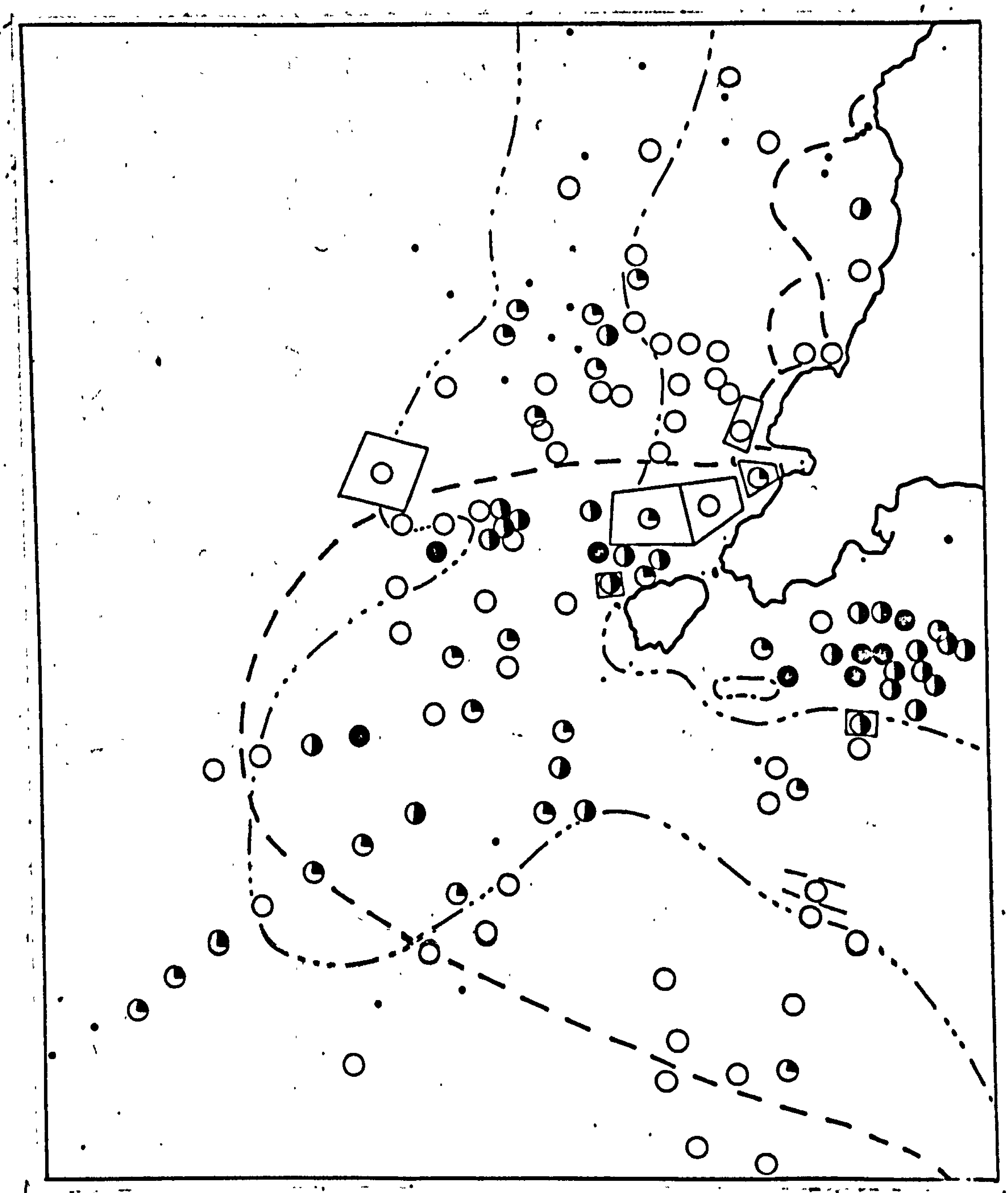
Map 53. Microporella ciliata. Relativeimportance in the ectoproct population within the area sampled.

• = locality at which M.ciliata was not obtained,
○ = locality at which less than 5 % ,
◔ = locality at which from 5 - 10 %,
◑ = locality at which from 10- 25 %,
● = locality at which more than 25% of the ectoprocts are M.ciliata, rest as Map 1.



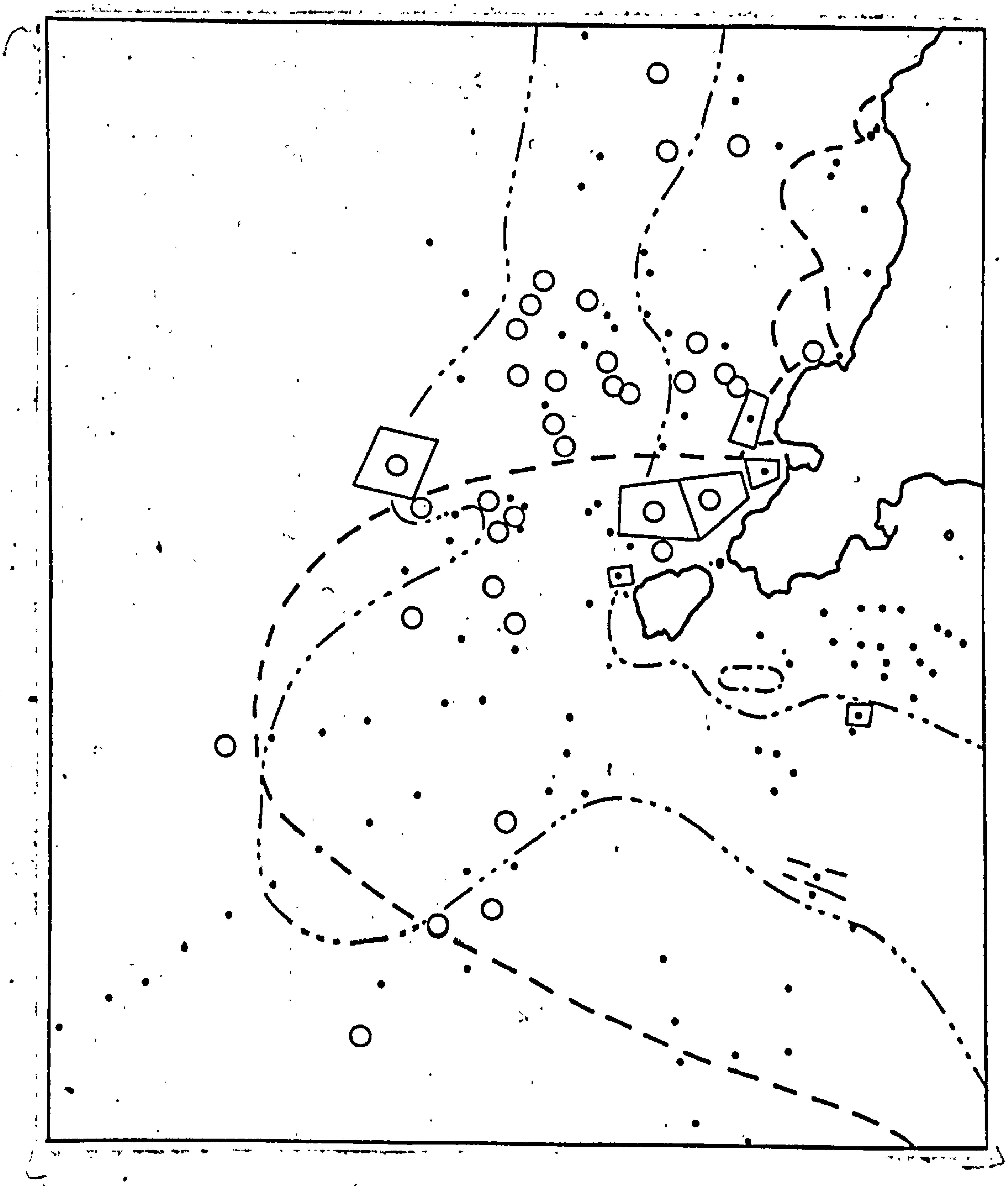
Map 54. Fenestrulina malusi. Distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

• = locality at which F. malusi was not obtained,
○ = less than 5 colonies per 1000 cm^2,
◔ = from 5 - 10 colonies per 1000 cm^2,
◑ = ffrom 10 - 25 colonies per 1000 cm^2,
◕ = from 25 - 50 colonies per 1000 cm^2,
● = more than 50 colonies per 1000 cm^2, rest as Map 1.



Map 55. Fenestrulina malusi. Relative importance in the ectoproct population within the area sampled.

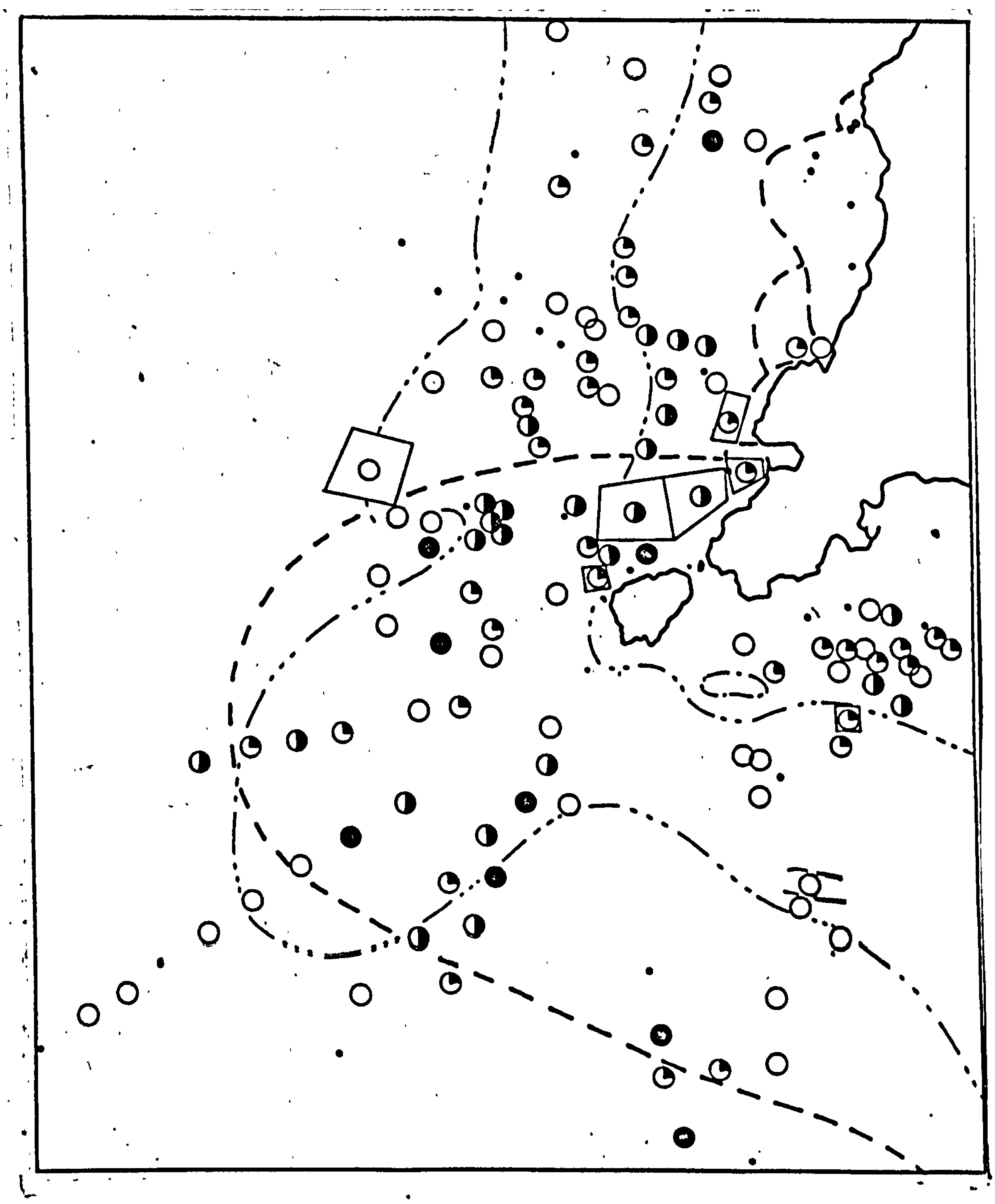
• = locality at which F.malusi was not obtained,
○ = locality at which less than 5 % ,
◔ = locality at which from 5 - 10 %,
◑ = locality at which from 10 - 20 %,
● = locality at which more than 20 % of the ectoprocts are F. malusi, rest as Map 1.



Map 56. Palmicellaria skenei. Distribution within the area sampled.

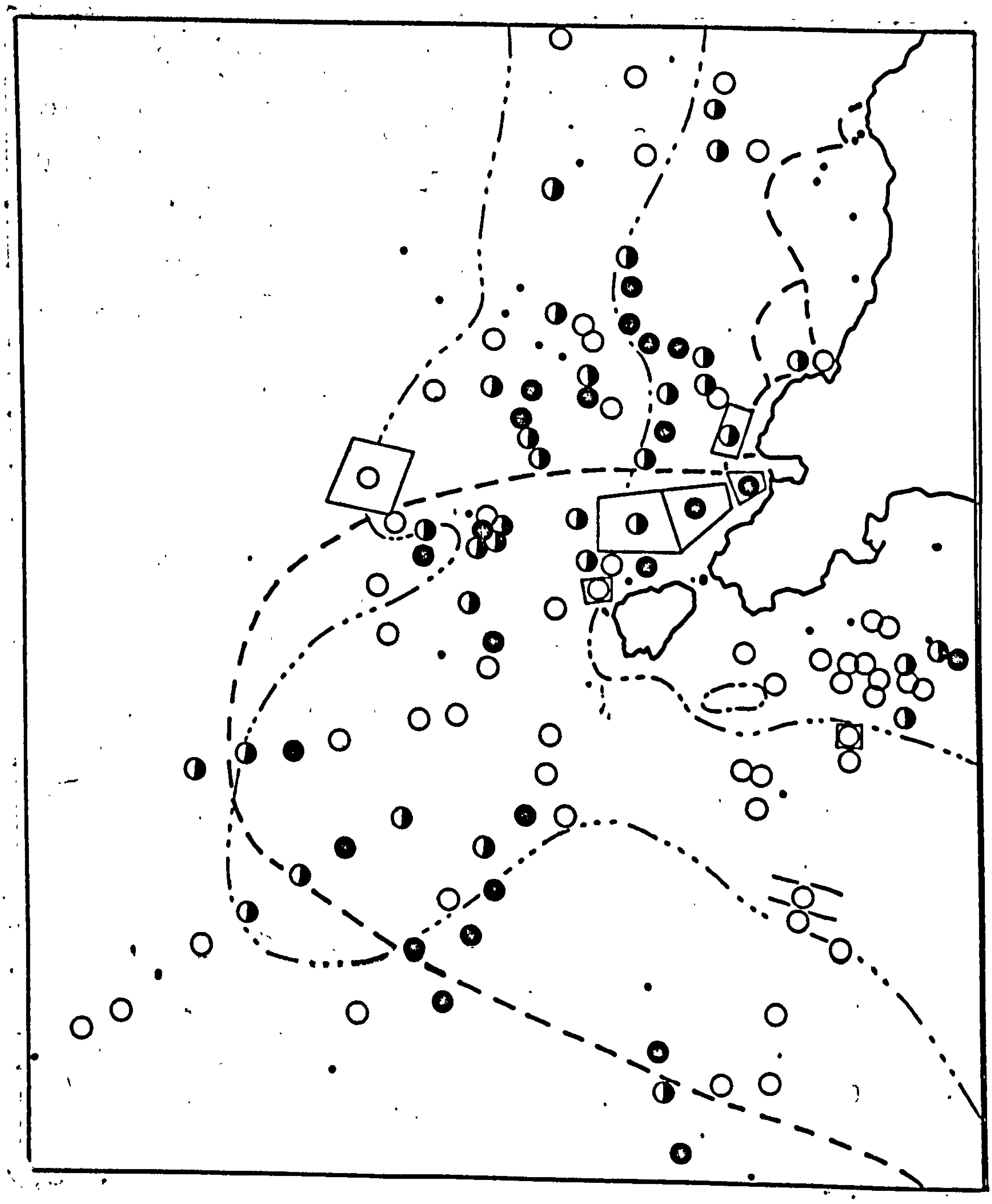
• = locality at which P.skenei was not obtained,

O = locality at which P.skenei was obtained., rest as Map 1.



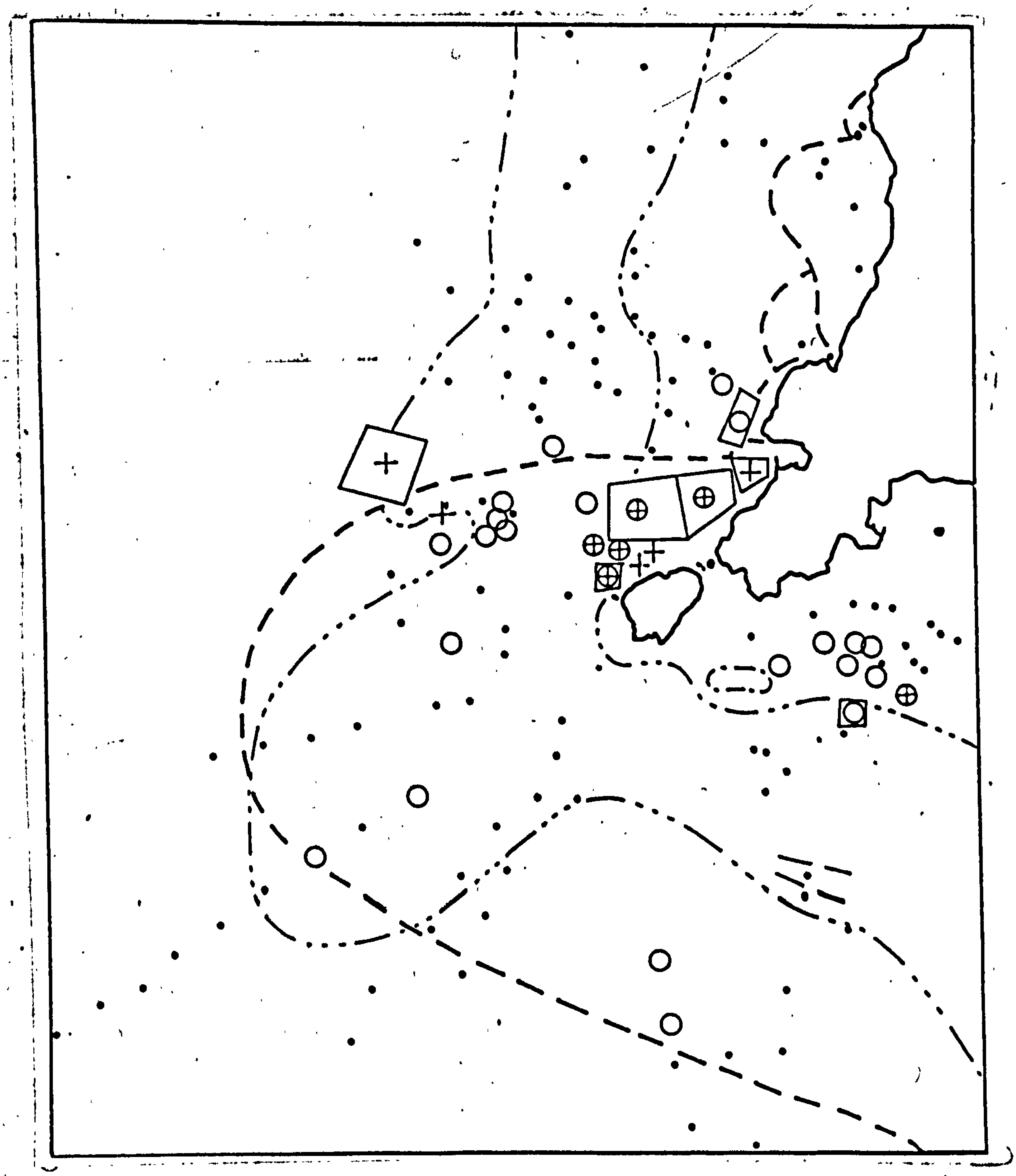
Map. 57. Porella concinna. Distribution and abundance within the area sample sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

- • = locality at which P.concinna was not obtained,
- ○ = less than 5 colonies per 1000 cm^2,
- ◔ = from 5 - 10 colonies per 1000 cm^2,
- ◑ = from 10 - 25 colonies per 1000 cm^2,
- ● = more than 25 colonies per 1000 cm^2, rest as Map 1.



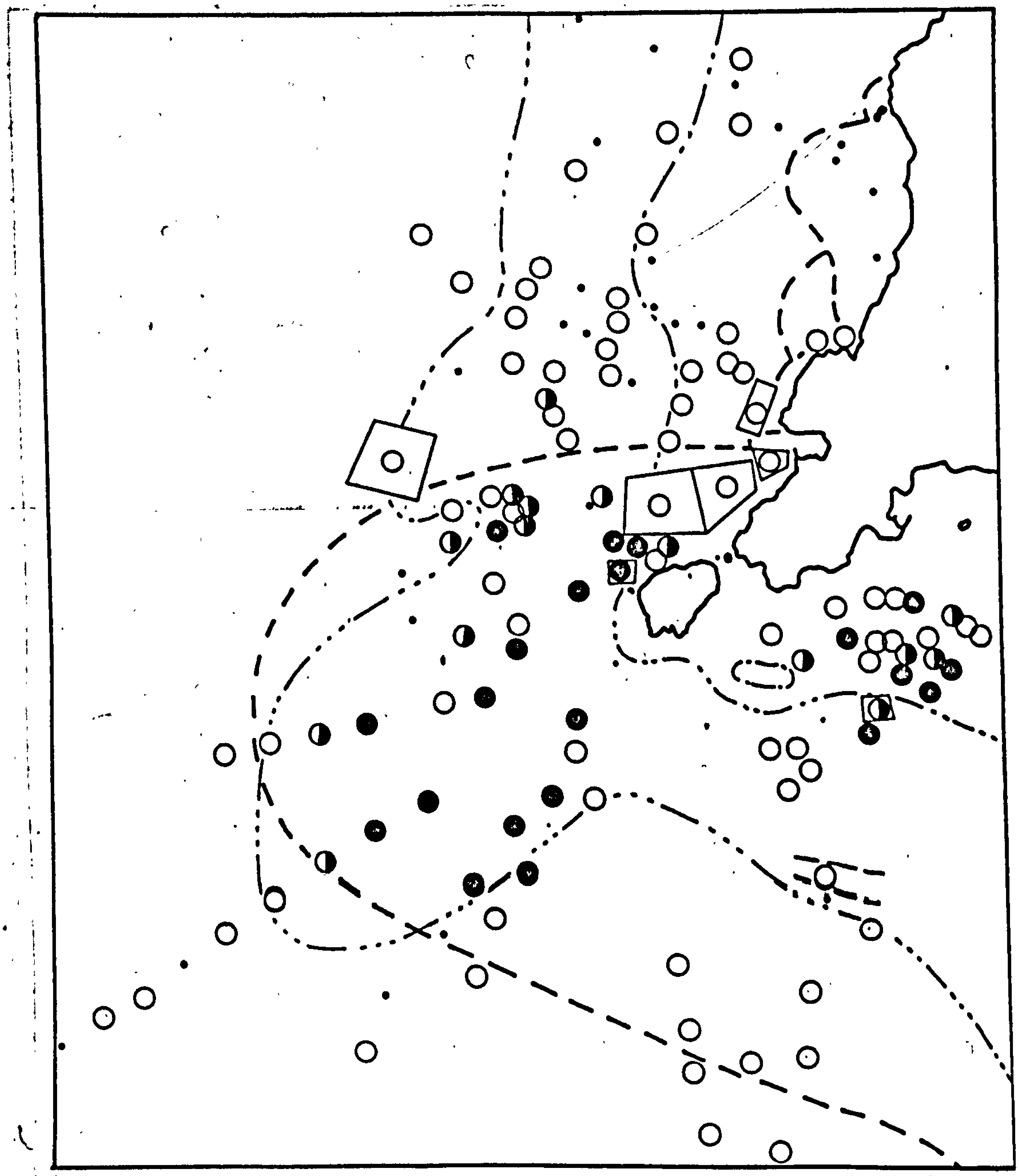
Map 58. Porella concinna. Relative importance in the ectoproct population within the area sampled.

• = locality at which P.concinna was not obtained,
○ = locality at which less than 5 %,
◐ = locality at which from 5 - 10 %,
● = locality at which more than 10% of the ectoproct colonies are Porella concinna, rest as Map 1.



Map 59. Smittina landsborovi and Smittina cheilostomata. Distribution within the area sampled.

+ = locality at which S.landsborovi was obtained,
O = locality at which S. cheilostomata was obtained,
• = locality at which neither was obtained., rest as Map 1.



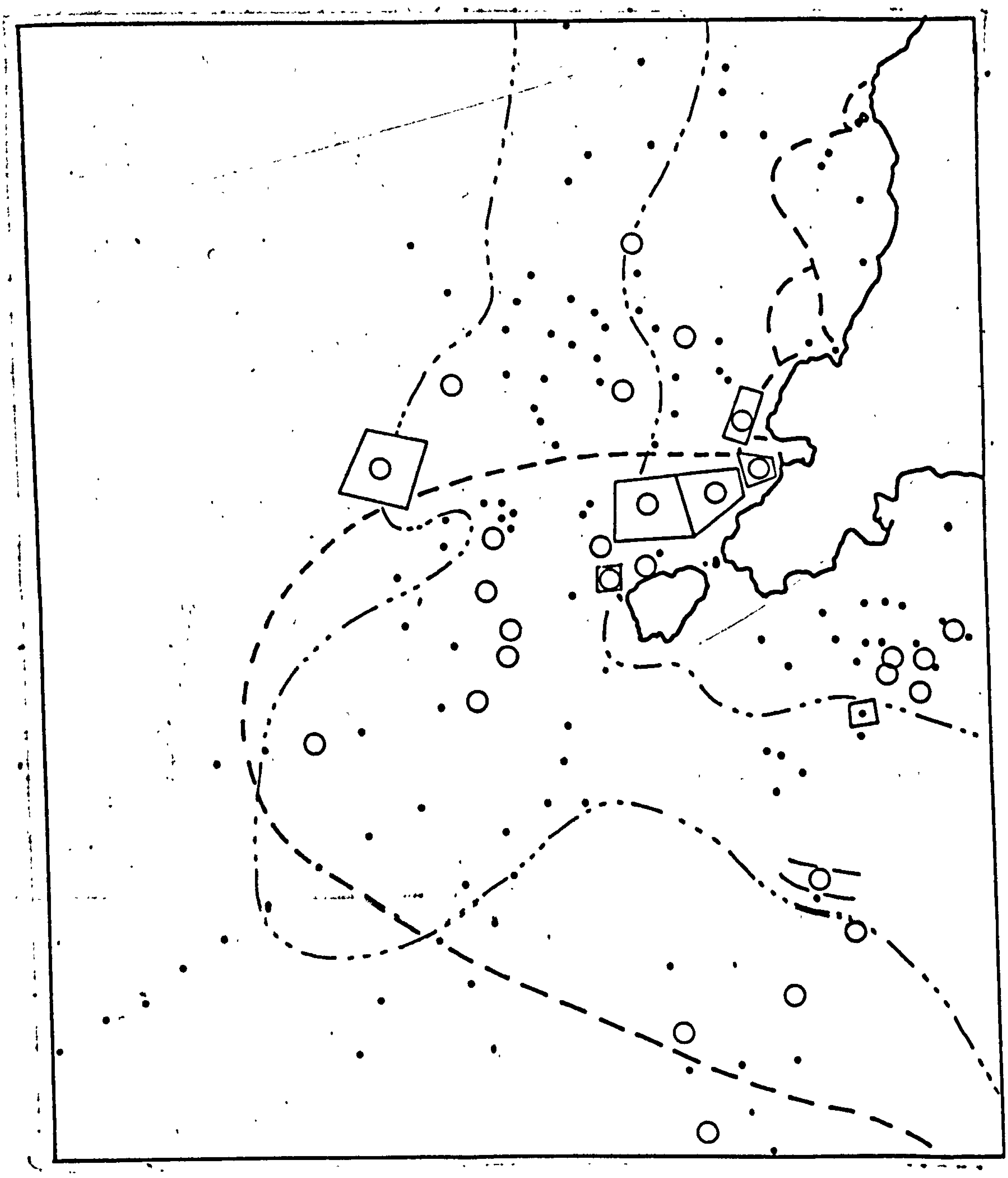
Map 60. Parasmittina trispinosa. Offshore distribution and abundance within the area sampled in terms of number of colonies per 1000 cm^2 surface area of support (excluding erect zoophytes) suitable for ectoproct colonization.

• = locality at which P. trispinosa was not obtained,

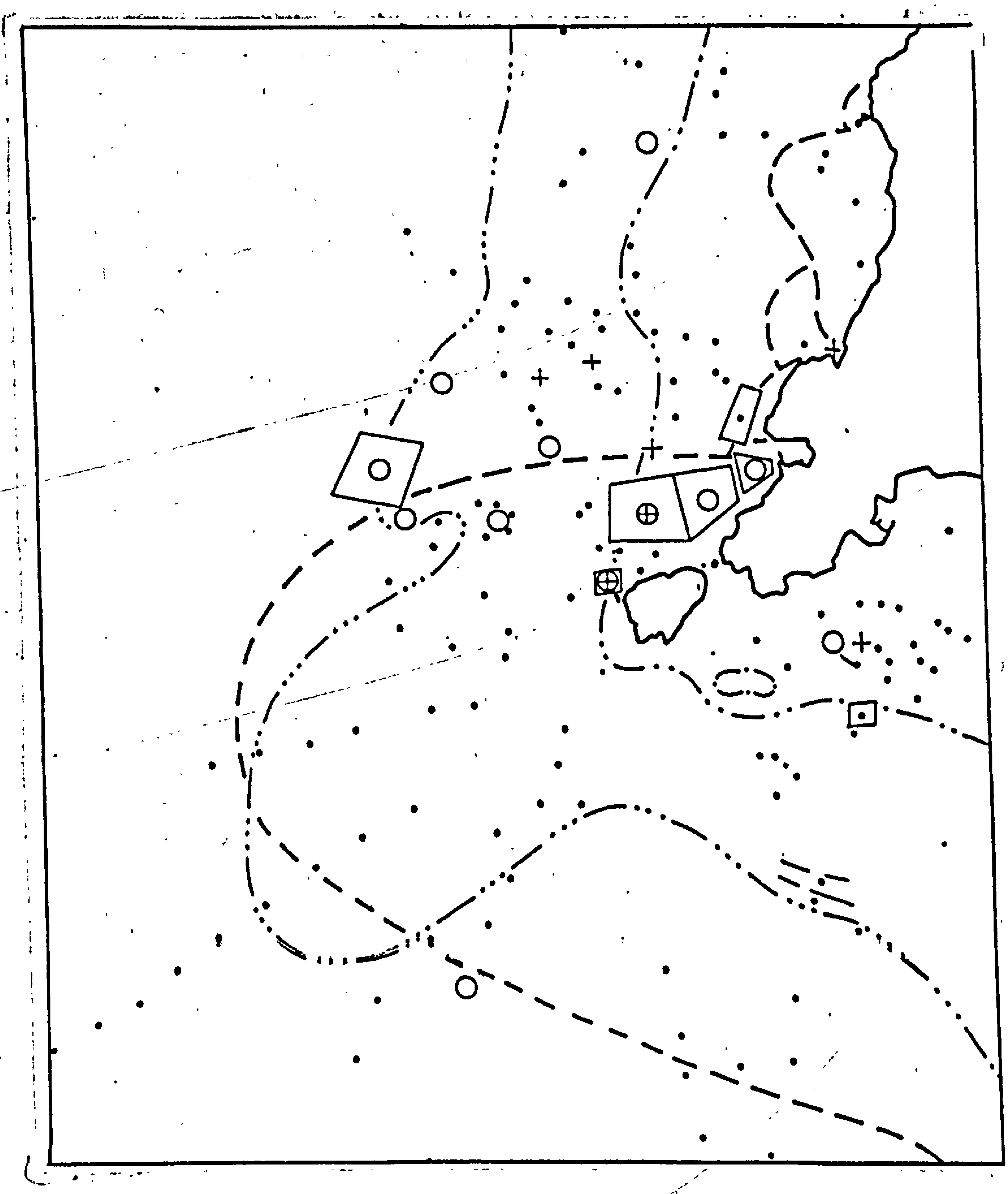
○ = less than 5 colonies per 1000 cm^2,

◑ = from 5 - 10 colonies per 1000 cm^2,

● = more than 10 colonies per 1000 cm^2, rest as Map 1.

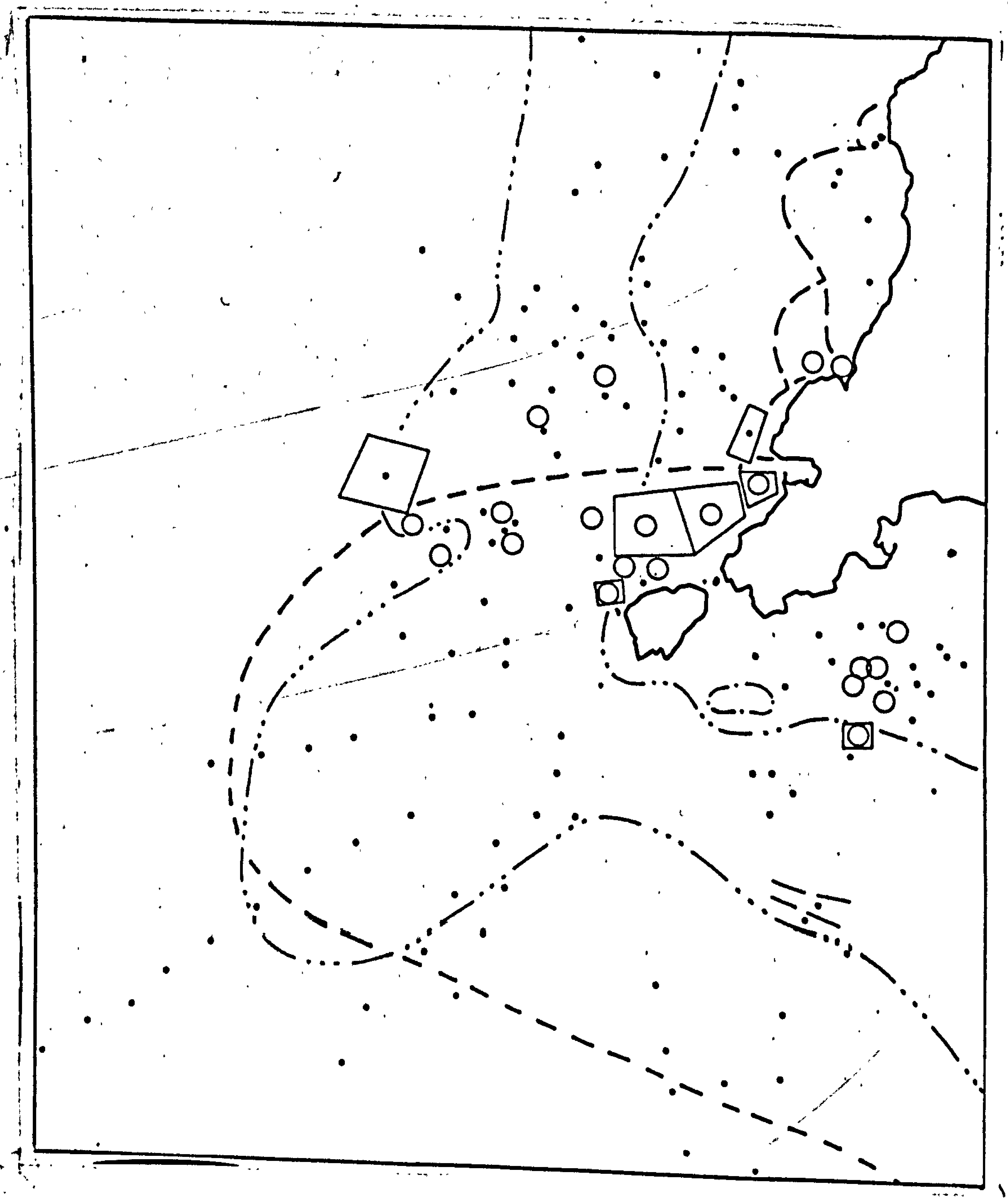


Map 61. Smittoidea reticulata. Distribution within the area sampled.
• = locality at which S.reticulata was not obtained,
O = locality at which S.reticulata was obtained, rest as Map 1.



Map 62. Phylactella collaris and Phylactella labrosa.
Distribution within the area sampled.

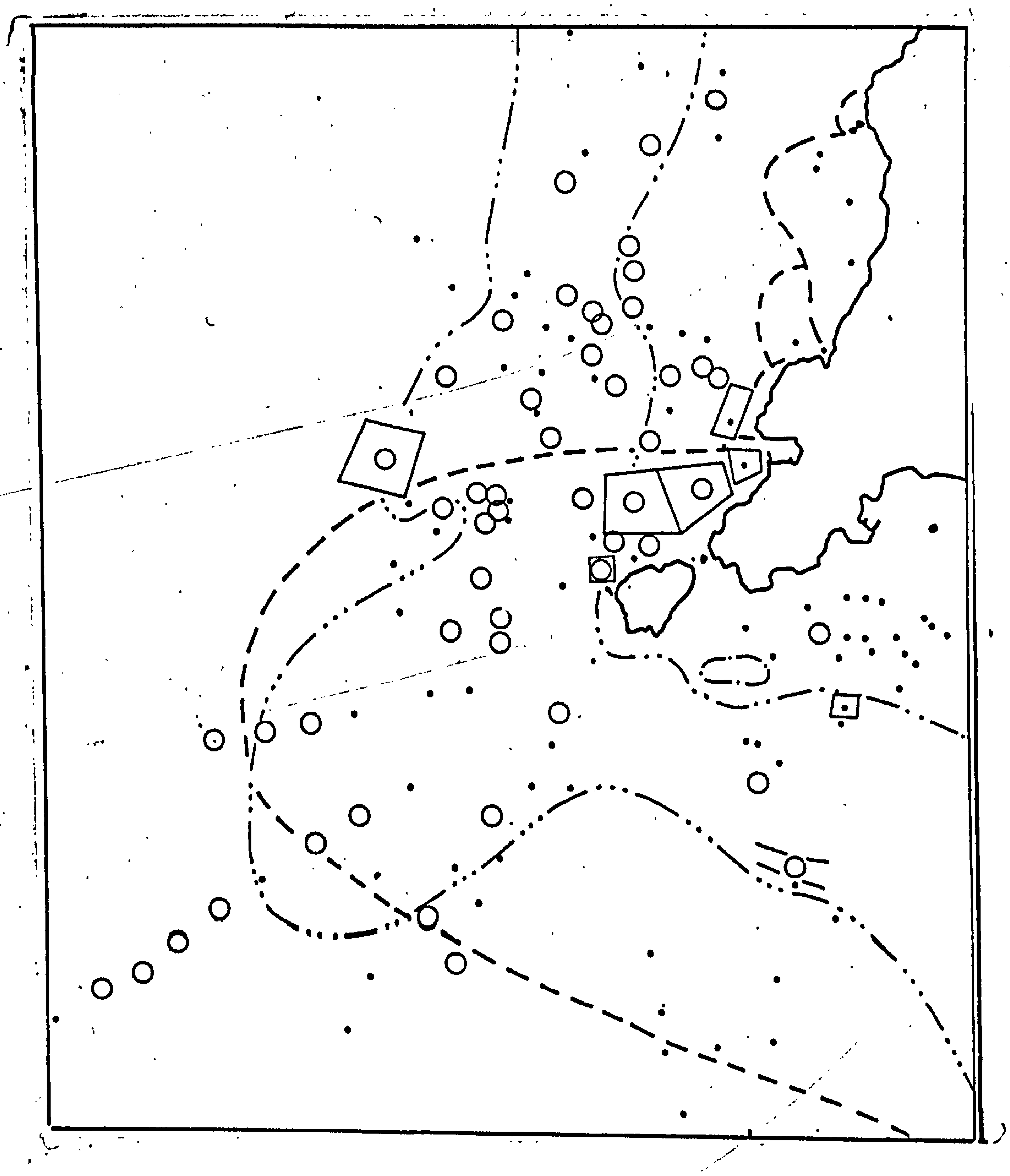
O = locality at which P. collaris was obtained,
+ = locality at which P. labrosa was obtained,
• = locality at which neither was obtained, rest as Map 1.



Map 63. Escharoides coccineus. Offshore distribution within the area sampled.

• = locality at which E.coccineus was not obtained,

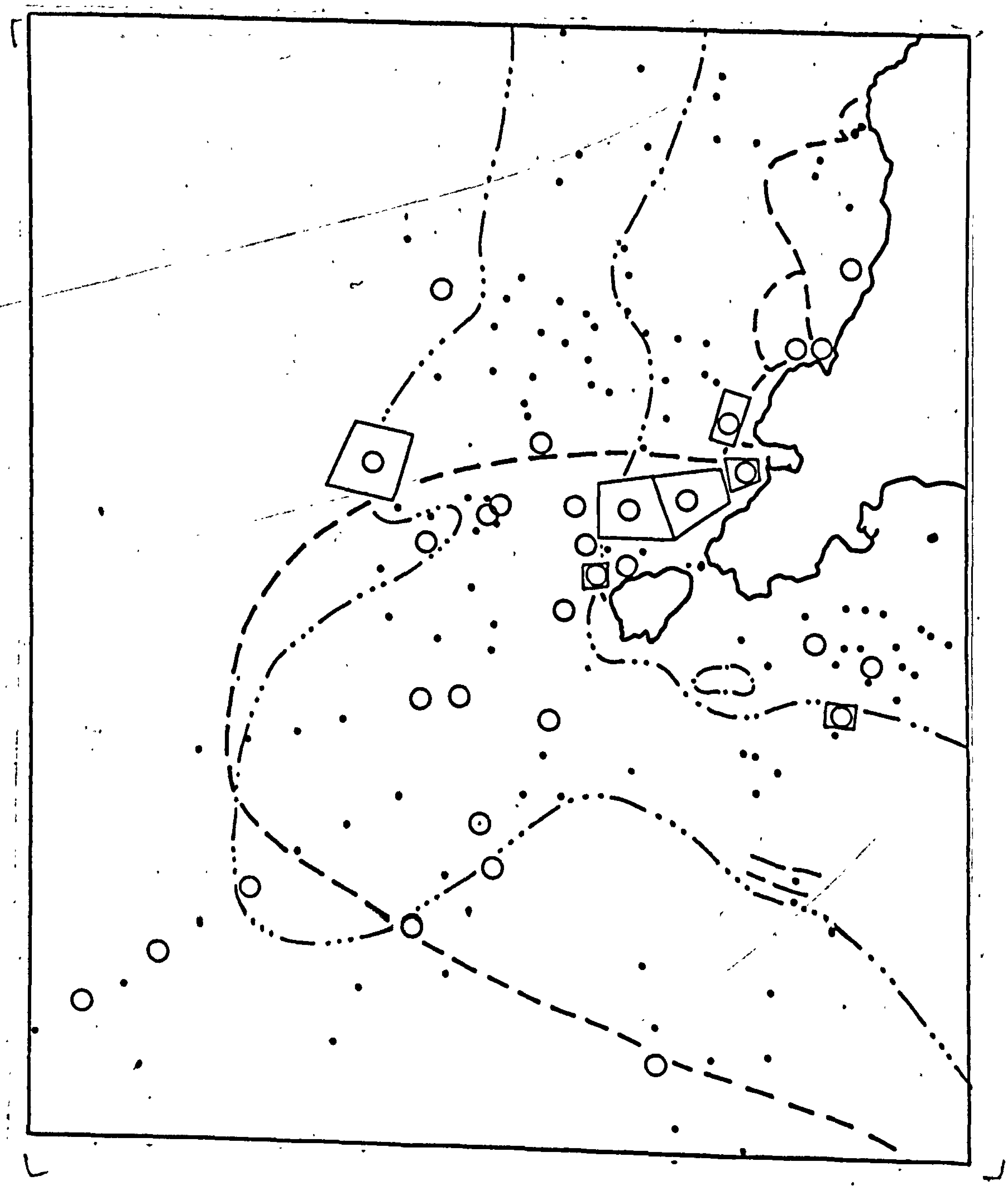
o = locality at which E.coccineus was obtained, rest as Map 1.



Map 64. Celleporaria dichotoma . Distribution within the area sampled.

• = locality at which C.dichotoma was not obtained,

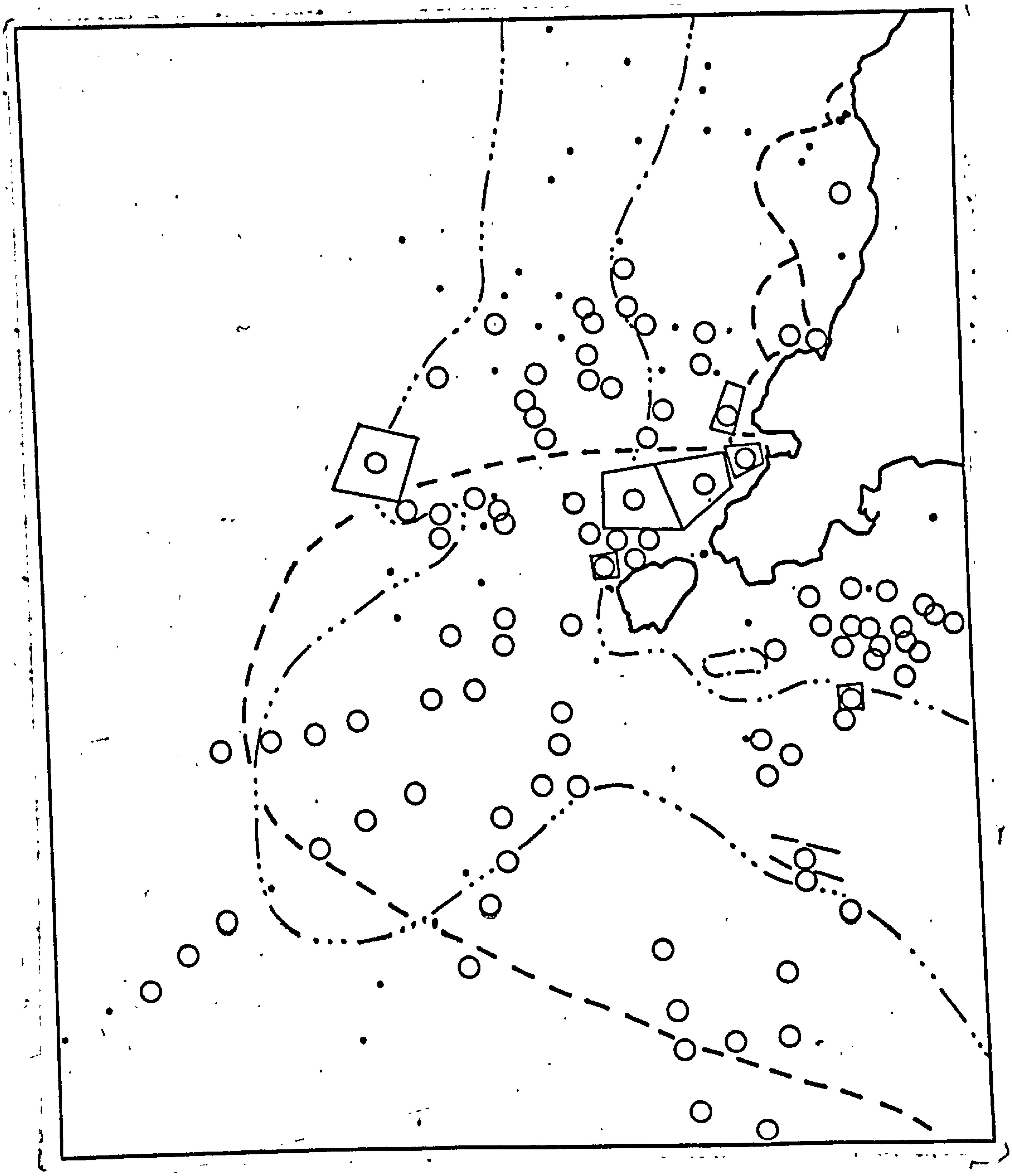
o = locality at which C.dichotoma was obtained, rest as Map 1.



Map 65. Celleporaria pumicosa. Offshore distribution within the area sampled.

• = locality at which C.pumicosa was not obtained,

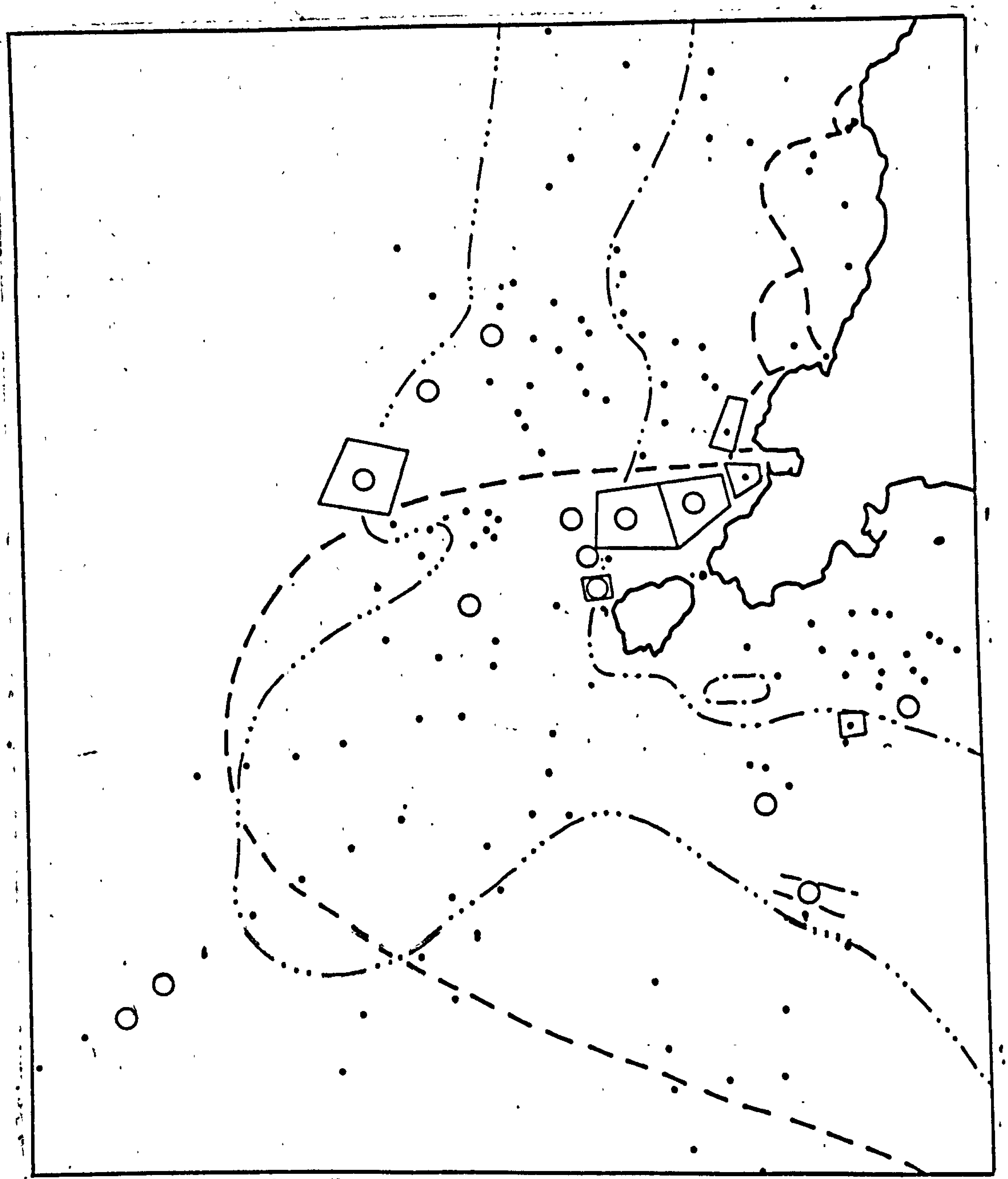
O = locality at which C.pumicosa was obtained, rest as Map 1.



Map 66. Osthimosia avicularis. Offshore distribution within the area sampled.

• = locality at which O.avicularis was obtained,

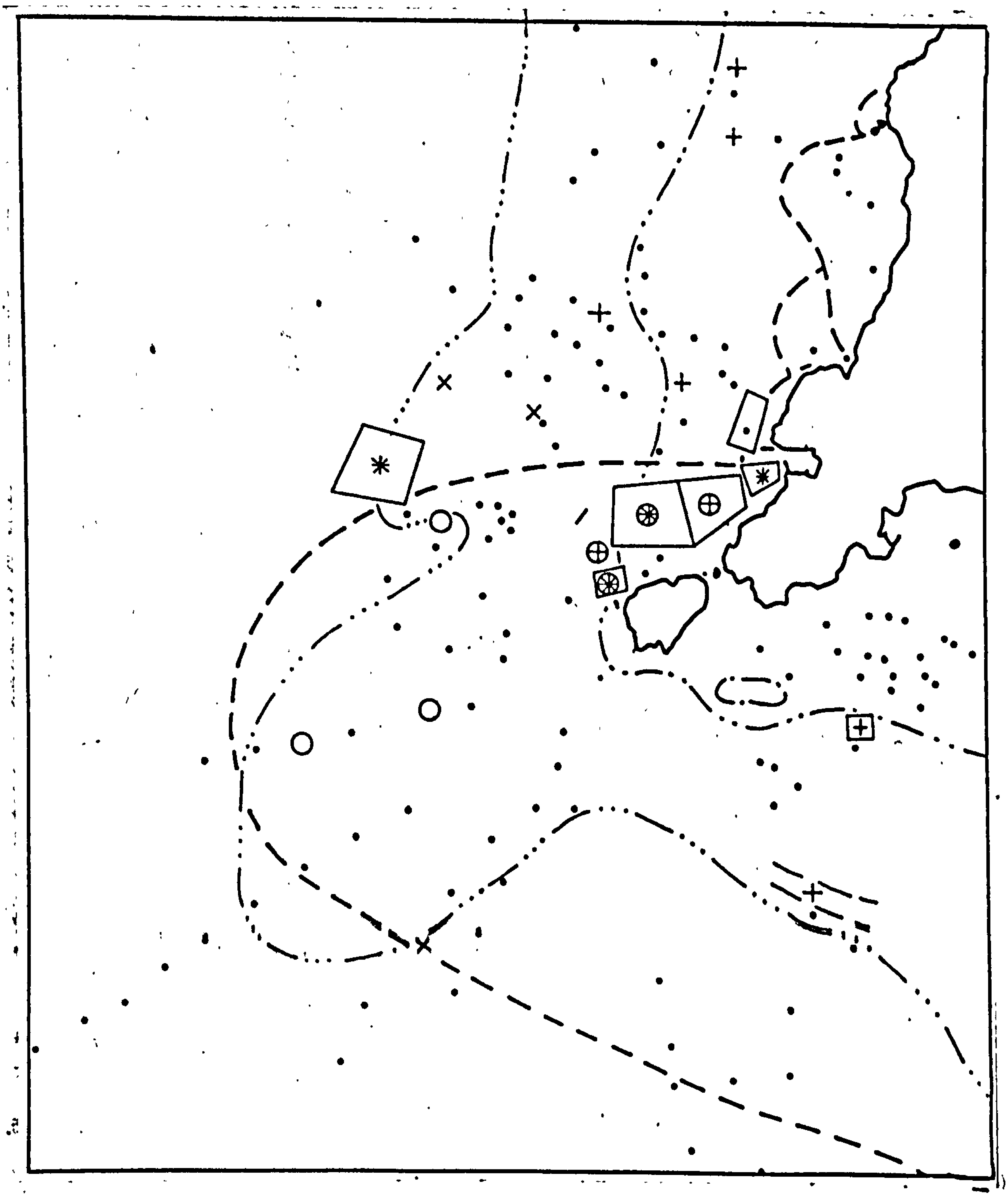
○ = locality at which O.avicularis was not obtained, rest as Map 1.



Map 67. <u>Celleporina</u> <u>costazii.</u> Offshore distribution within the area sampled.

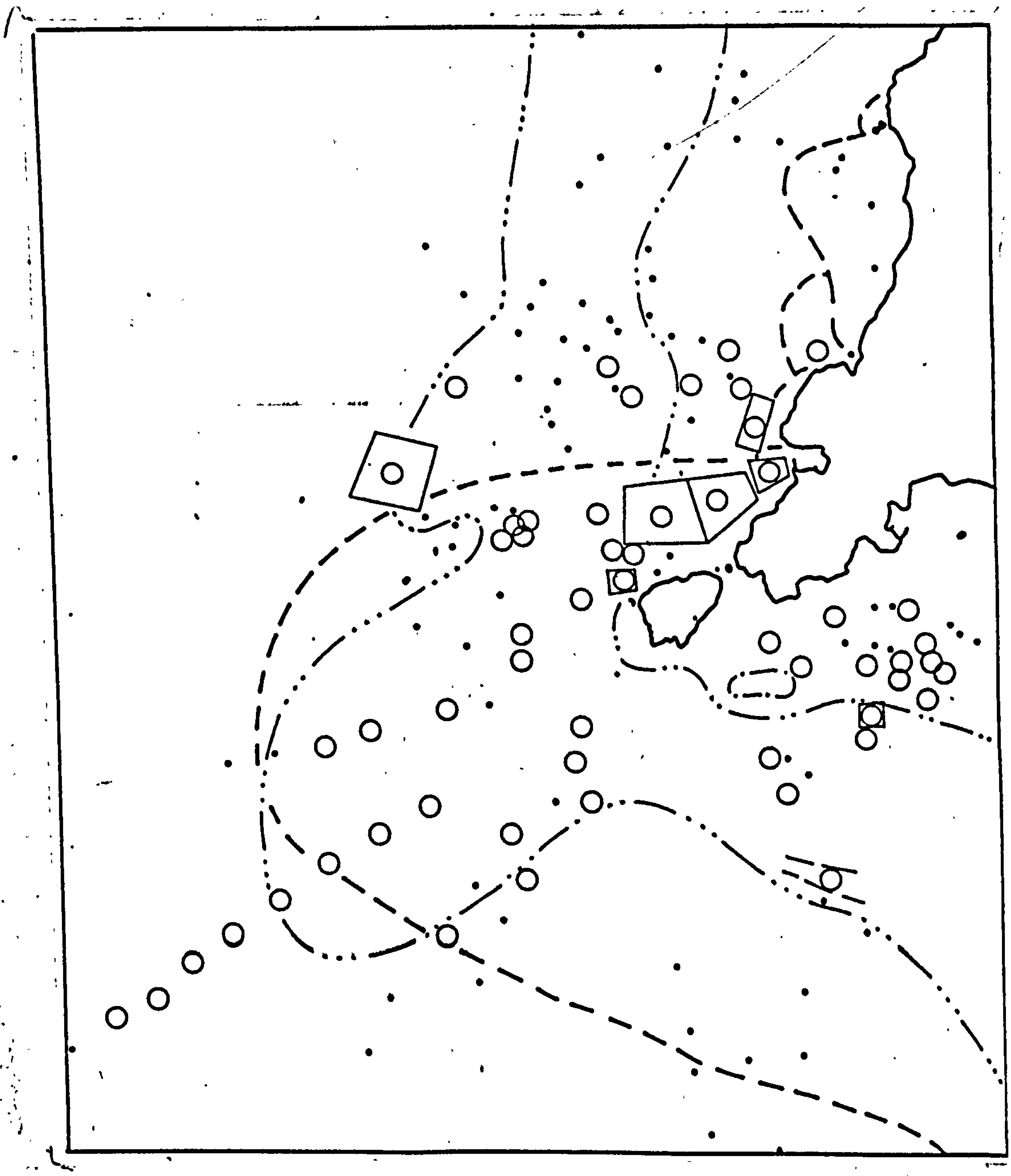
• = locality at which <u>C.costazii</u> was not obtained,

○ = locality at which <u>C.costazii</u> was obtained, rest as Map 1.



Map 68. Alcyonidium mamillatum, Alcyonidium gelatinosum, and Alcyonidium variegatum. Offshore distribution within the area sampled.

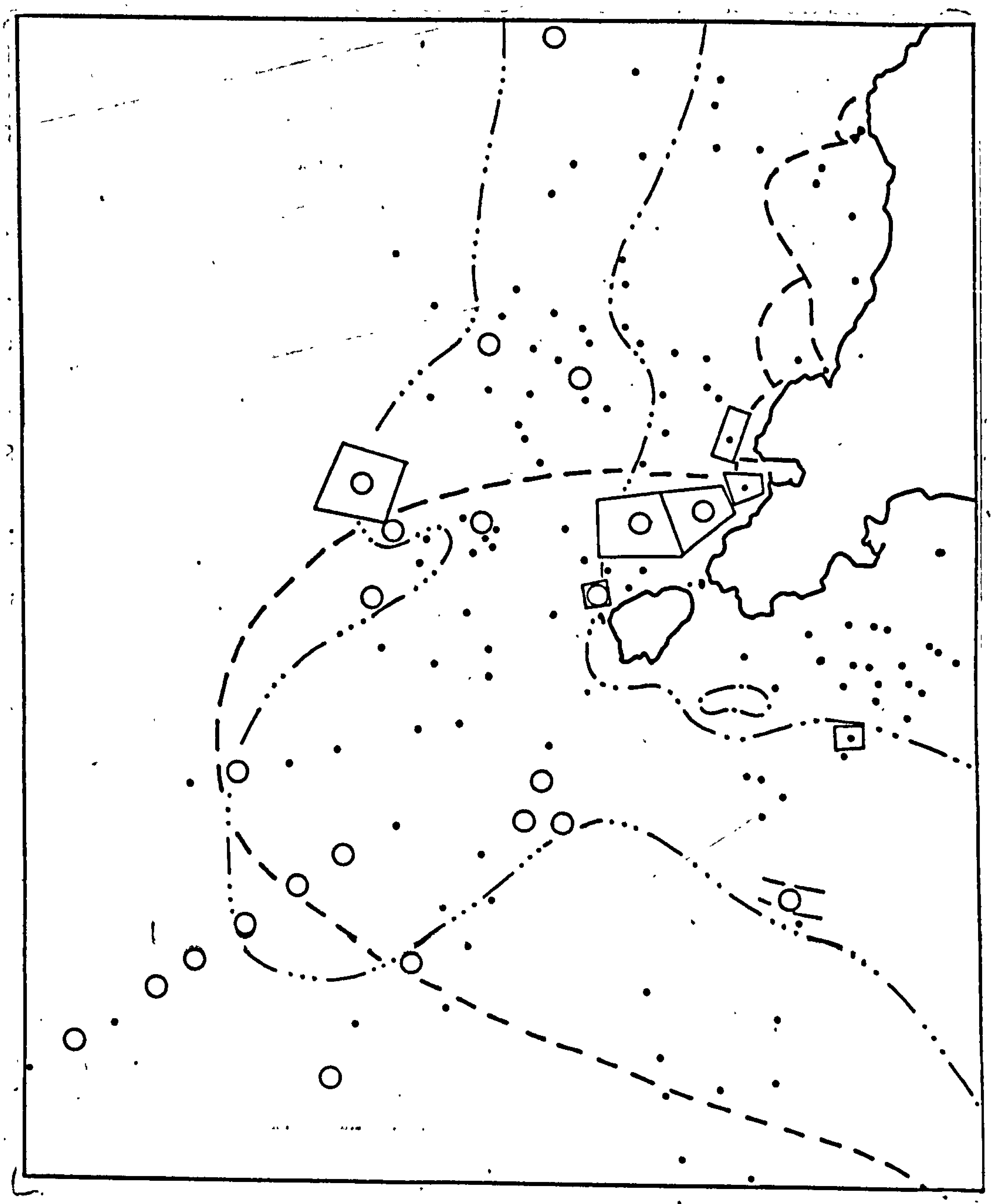
o = locality at which A. mamillatum was obtained,
+ = locality at which A. gelatinosum was obtained,
x = locality at which A. variegatum was obtained,
• = locality at which none of these species was obtained,
rest as Map 1.



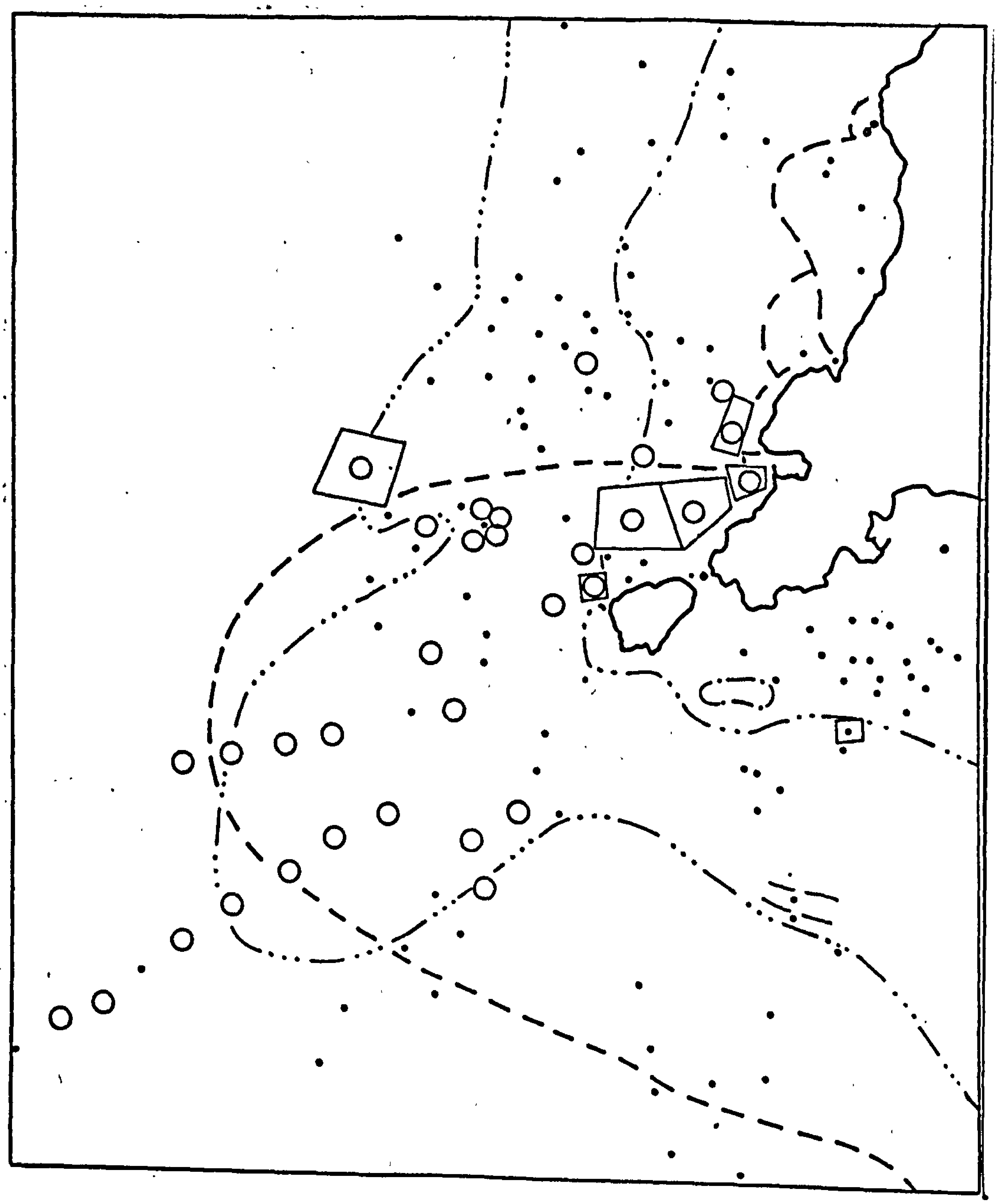
Map 69. Alcyonidium mytili. Offshore distribution within the area sampled.

• = locality at which A.mytili was not obtained,

o = locality at which A.mytili was obtained, rest as Map 1.



Map 70. Arachnidium hippothooides. Distribution within the area samplcd.
• = locality at which A.hippothooides was not obtained,
○ = locality at which A.hippothooides was obtained,
rest as Map 1.



Map 71. Nolella dilatata. Distribution within the area sampled.
O = locality at which N.dilatata was obtained,
• = locality at which N.dilatata was not obtained, rest as Map 1.

Appendices

Appendix I. Sampling Stations. (See Map 1)

Sample	Locality (Statute miles; True bearings)			Depth (fathoms)	Dredge Contents (ch = shell st= stone gr = gravel z = zoophytes)
1.	4·6 ml	W35° N	of Niarbyl	24	st. ch.
2.	3·2 ml	W21° N	"	22	ch. Luidia, Echinus.
3.	2·2 ml	W26° N	"	17	st. ch. Pecten
4.	2·1 ml	W14° N	"	22	ch.st. Pecten. Ophiura.
5.	4·2 ml	W2° S	"	24	Empty
6.	4·5 ml	W7·5° S	"	25	ch. Pecten, Porania, Cyprina.
7.	3·1 ml	W2° S	"	21	ch. Pecten.
8.	2·0 ml	W	"	19	st. ch. Luidia? Pecten
9.	·9 ml	W6° S	"	18	ch. Pecten
10.	·6 ml	W40° S	"	16	Empty
11.	·8 ml	W40° S	"	16	St. ch. Pecten, Asterias.
12.	1·1 ml	S	"	12	gr. ch. Glycymeris, Astropecten.
13.	2·1 ml	S	"	16	ch. Pecten, Lutraria, Asterias, Solaster.
14.	4·0 ml	W30° S	"	20	ch, large st? Chlamys Pecten, Ophiothrix.
15.	3·9 ml	W16° S	"	18–13	ch-gr. ch. z. Chlamys Glycymeris? [illegible].
16.	5·2 ml	N28° W	of Thousla	27	ch. Cyprina? Palmipes.
17.	6·6 ml	NW	"	34	Ch. z. Cyprina.
18.	5·7 ml	NW	"	26	Pecten, Cyprina, ch. Dentalium.
19.	5·0 ml	N34° W	"	30	ch. Cyprina, Echinus
20.	4·0 ml	N54° W	of Breakwater Bouy (Port Erin)	24	st.ch. Pecten
21.	3·7 ml	N50° W	"	23	ch. Cyprina, Pecten Astropecten, Luidia
22.	3·4 ml	N54° W	"	22	ch. Cyprina, Pecten.
23.	3·2 ml	N43° W	"	23	ch, st. Pecten, Cyprina Porania.
24.	2·6 ml	N37° W	"	22–23	ch. Pecten, Chlamys, Cyprina, Palmipes.
25.	3·6 ml	N3° W	of Thousla	20	ch. st. Pecten, Palmipes Porania.
26.	2·0 ml	N29° W	of Breakwater Bouy (Port Erin)	21	Sh. st. Palmipes, Cyprina
27.	2·7 ml	N5° E	"	18–20	ch. st. Cyprina, Pecten

Sample	Locality			Depth	Dredge Contents.
28.			Fleshwick Bay	15-10	st. Pecten, Henricia, Asterias.
29.	1·4 ml	N31° W	of Breakwater Bouy (Port Erin)	22	sh. Pecten, Asterias, Luidia Palmipes, Porania
30.	1·5 ml	N30° W	"	19	sh. Pecten, Asterias, Palmipes.
31.	2·0 ml	N54° W	"	22	sh. Pecten Asterias
32.	4·3 ml	N	of Chickens	19	sh. Chlamys Pecten, Cyprina Scaphander
33.	2·7 ml	N65° W	of Breakwater Bouy (Port Erin)	27	sh. Pecten, Glycymeris z.
34.	3·2 ml	N62° W	"	25	sh. z. Pecten, Chlamys Buccinum
35.	3·6 ml	N60° W	"	36	Empty
36.	4·0 ml	N60° W	"	38-33	Empty
37.	4·7 ml	N39° W	of Thousla	26	Sh. Pecten Scaphander Cyprina Chlamys
38.	5·3 ml	N27° W	of Chickens	32	sh. z. Aporrhais.
39.	5·0 ml	N26° W	"	30	sh. z. Cancer.
40.	4·2 ml	N44° W	of Thousla	25	sh. Cyprina, Pecten
41.	4·8 ml	N50° W	of Breakwater Bouy (Port Erin)	24	sh. Cyprina Pecten Astropecten, Ophiura.
42.	3·3 ml	N47W	of Thousla	23	sh. z. st. Pecten
43.	3·4 ml	N86° W	of Breakwater Bouy (Port Erin)	27	sh. z. Pecten Henricia
44.	4·2 ml	N13° W	of Chickens	20-21	sh. Pecten Porania
45.	3·8 ml	N72° W	of Breakwater Bouy (Port Erin)	25	st. sh. Pecten, Echinus Astropecten
46.	2·1 ml	N23W	of Thousla	22	sh. z. Pecten, Chlamys, Glycymeris.
47.	2·4 ml	N8° W	"	20	Sh. Pecten Echinus Porania Palmipes
48.	2·0 ml	N58° W	"	22	Pecten, Chlamys, z. sh.
49.	2·0 ml	N65° W	"	19	sh. z. Chlamys, Pecten Echinus, Asterias
50.	2·6 ml	N32° W	of Chickens	27	st. sh. Pecten, Asterias
51.	2·8 ml	N32° W	"	30	sh. z. Pecten, Asterias Ophiothrix, Asterias
52.	2·8 ml	N29° W	"	27	st. sh. Echinus Asterias
53.	3·0 ml	N31° W	"	27	sh. Palmipes, Porania.
54.	3·2 ml	N35° W	"	34	sh. z.
55.	5·2 ml	S79° W	of Breakwater Bouy (Port Erin)	32	sh. Pecten, Cancer, Scaphander Hermione
56.	3·8 ml	N51° W	of Chickens	40	Pecten, Dentalium sh.
57.	3·5 ml	N66° W	"	42-44	sh. Buccinum Eupagurus Aporrhais.
58.	3·2 ml	N52° W	"	39	sh. gr. Pecten Palmipes Eupagurus
59.	2·7 ml	N39° W	"	27	sh. sh-gr. Pecten Chlamys
60.	1·8 ml	N	"	18-20	sh. Chlamys Henricia.

Sample	Locality			Depth	Dredge Contents
61.	1.8ml	N9° E	of Chickens	27-23	sh.s.Cancer,Eledone Eupagurus
62.	.9 ml.	N87° W	of Thousla	16	st.Pecten
63.	.6 ml.	N67° W	"	21	sh.Pecten,Eledone, st.s.Echinus.
64.	1.4 ml.	N21° W	of Chickens	24	Large st.sh.Echinus Asterias
65.	2.2 ml.	N56° W	"	32	sh.Pecten,Chlamys, Buccinum
66.	1.6 ml.	N65° W	"	27	sh.st.Spatangus, Asterias,Modiolus
67.	1.5 ml.	N79° W	"	28	sh.st.Solaster, Echinus.
68.	2.3 ml .	N79° W	"	24	st.sh.s. Pecten Buccinum
69.	2.7 ml.	N74° W	"	34	sh.Dentalium
70.	3.2 ml.	N75° W	"	35	sh.Pecten,Cyprina, Colus
71.	2.0 ml.	S79° W	"	23	st.sh.Echinus, Asterias
72.	2.6 ml.	S79° W	"	27	sh.Pecten,Modiolus Echinus, Spatangus
73.	3.7 ml.	S79° W	"	27	large st. sh.Chlamys Pecten
74.	4.6 ml.	S79° W	"	27	sh.sh-gr.Chlamys, Pecten,Gari
75.	5.4 ml.	S79° W	"	30	sh.Pecten,Chlamys Buccinum
76.	6.1 ml.	S79° W	"	35	sh.st.Pecten,Asterias, SolasterAstropecten
77.	3.6 ml.	S57° W	"	27	st. sh.Asterias.
78.	4.5 ml.	S57° W	"	23	sh.Spatangus, Henricia
79.	5.4 ml.	S57° W	"	27	st.sh.Pecten,Chlamys, Colus,Hermione.
80.	6.3 ml.	S 57° W	"	30	sh.Pecten,Chlamys, Colus,Hermione,
81.	7.2 ml.	S57° W	"	33	sh.Pecten,Colus
82.	8.0 ml.	S57° W	"	30	sh.Astropecten, Pecten
83.	8.9 ml.	S57° W	"	30	Asterias
84.	9.4 ml.	S57° W	"	33	sh.Alcyonium,Pecten, Asterias,Colus,Luidia
85.	10.3 ml.	S57° W	"	36-40	Cyprina,Pecten Asterias(only)
86.	6.9 ml.	S34° W	"	32	Chlamys Pecten,Cyprina Spatangus
87.	5.9 ml.	S34° W	"	32	Chlamys, Pecten, Buccinum,Asterias
88.	4.8 ml.	S34° W	"	33	sh.Asterias,Buccinum Eupagurus Ophiura
89.	3.9 ml.	S34° W	"	30	sh,Asterias, Eupagurus
90.	2.9 ml.	S34° W	"	28	sh.Asterias, Eupagurus
91.	1.9 ml.	S34° W	"	30	sh.sh-gr. Buccinum Modiolus,Chlamys
92.	.9 ml.	S34° W	"	27	st.sh.Alcyonium, Luidia
93.	1.5 ml.	S25° W	"	32	sh.sh-gr.Buccinum Spatangus

Sample	Locality			Depth	Dredge Contents
94.	2·8 ml.	S12° W	of Chickens	32	sh.s. Modiolus, Buccinum, Clinker, Asterias, Solaster
95.	2·2 ml.	S25° W	"	32-36	sh. Spatangus, Palmipes, Porania, Solaster
96.	3·4 ml.	S25° W	"	30	sh, large st; Pecten, Echinus Asterias
97.	4·2 ml.	S25° W	"	32-38	large st. ?Pecten, Buccinum, Eupagurus
98.	5·2 ml.	S25° W	"	35	sh. Pecten Chlamys Buccinum Palmipes Porania
99.	4·5 ml.	S11° E	"	28	sh. Modiolus, Alcyonium Tritonia
100.	5·5 ml.	S11° E	"	37	sh. Spatangus, Ophiuroids
101.	6·1 ml.	S11° E	"	37	st. sh. Spatangus, Echinus Palmipes
102.	7·1 ml.	S11° E	"	37 5	sh. Chlamys, Buccinum, Echinus Spatangus, Palmipes, Porania
103.	7·6 ml.	S20° E	"	37	St. sh. Chlamys, Asterias, Ophiuroids.
104.	6.2 ml.	S19° E	"	37	Ophiuroids
105.	6·4 ml.	S25° E	"	37	Ophiuroids, Chlamys, Spatangus
106.	5·6 ml.	S30° E	"	37	sh. s. Asterias, Buccinum, Modiolus
107.	5·4 ml.	S54° E	"	33	Ophiuroids, st. sh. Modiolus
108.	4·7 ml.	S81° E	"	32	Ophiuroids, Modiolus, st. sh. Tritonia, Buccinum.
109.	3·2 ml.	S52° E	"	32	Ophiuroids, st. sh. Asterias
110.	3·2 ml.	S62° E	"	32	st. sh. Glycymeris, Modiolus
111.	2·7 ml.	S61° E	"	25-26	st. s. Alcyonium, Asterias, Echinus
112.	2·8 ml.	S65° E	"	27	st. Ophiuroids, Asterias Glycymeris
113.	3·9 ml.	S76° E	"	23-24	sh. Pecten, Asterias, Echinus
114.	2·4 ml.	N79° E	"	20	Ophiuroids, sh.
115.	2·7 ml.	N88° E	"	22	sh-gr. sh. Pecten, Buccinum.
116.	3·4 ml.	N75° E	"	20	Ophiuroids. sh. large st. Alcyonium.
117.	4·0 ml.	N75° E	"	18	Ophiuroids, st. sh.
118.	4·3 ml.	N77° E	"	18	Ophiuroids, Glycymeris, sh. Echinus, Asterias.
119.	4·6 ml.	N79° E	"	17	gr. coralline-gr. sh. Echinus Spatangus, Asterias
120.	4·2 ml.	S 87° E	"	20	Ophiuroids, sh. Echinus,
121.	3·9 ml.	N86° E	"	16-18	sh-gr. sh. Pecten, Chlamys.
122.	4·2 ml.	N86° E	"	18	sh. Chlamys
123.	4·7 ml.	N86° E	"	18	sh. Asterias, Porania, Echinus
124.	5·1 ml.	N83° E	"	20	Ophiuroids, sh.

Sample		Locality		Depth	Dredge Contents
125.	5.2 ml.	N84°E	of Chickens	17	large st.Asterias,Echinus Porania
126.	5.5 ml.	N86°E	"	17	Ophiuroids,sh.st.Echinus Asterias
127.	5.0 ml.	E	"	19	sh.Echinus,Asterias
128.	4.8 ml.	N89°E	"	21	sh.Asterias
129.	4.3 ml.	N89°E	"	22	sh.Asterias
130.	4.2 ml.	S87°E	"	20	sh.Pecten,Buccinum
131.	3.8 ml.	S88°E	"	20	Asterias,sh.Echinus Ophiuroids.
132.	4.6 ml.	S86°E	"	20	Asterias,Pecten,sh.

A

Appendix II . Records of reproduction (based only on presence of eggs of embryos) for Manx ectoprocts. Columns Nor. and Med. indicate wether or not each species has been recorded from Norway and the Mediterranean respectively. + positive record.

Species	Month												Distribution	
	J	F	M	A	M	J	J	A	S	O	N	D	Nor.	Med.
Crisidia cornuta	—	—	—	+	+	+	+	—	—	+	—	—	+	+
Crisia eburnea	—	—	—	+	+	+	—	—	—	—	—	—	+	+
Crisia aculeata	+	—	—	—	+	—	+	—	—	+	—	++	+	+
Crisia ramosa	+	—	+	—	—	—	—	+	+	+	+	+	—	+
Tubulipora phalangea	+	—	+	+	—	+	+	+	+	+	+	+	?	?
Tubulipora penicillata	—	—	—	—	—	—	—	—	+	—	—	—	+	+
Tubulipora lobulata	—	—	—	—	—	+	—	—	—	—	+	—	?	?
Diaperoecia major	—	+	+	—	—	+	—	—	—	—	—	—	+	+
Diplosolen obelia	—	—	—	+	—	—	—	—	—	—	—	—	+	+
Diastopora patina	—	—	—	—	—	+	+	+	—	—	—	—	+	+
Diastopora suborbicularis	+	+	+	+	—	—	—	—	+	+	—	+	+	+
Lichenopora hispida	—	+	+	+	—	+	—	—	+	—	—	—	+	+
Aetea anguinea	+	÷	÷	÷	÷	—	÷	+	—	—	—	—	+	+
Aetea sica	—	—	—	—	+	+	+	+	+	+	+	—	+	+
Eucratea loricata	—	—	+	—	—	—	—	—	—	—	—	—	+	+
Membranipora membranacea	—	—	—	+	+	+	—	—	—	—	—	—	+	+
Alderina imbellis	+	+	+	+	+	+	+	+	:	+	+	+	+	—
Callopora lineata	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Callopora dumerili	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Callopora aurita	+	+	—	+	—	—	—	+	+	+	+	+	+	—
Callopora craticula	+	—	+	+	+	—	+	+	+	+	+	+	+	—
Amphiblestrum flemingi	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Cauloramphus spiniferum	+	+	+	+	+	—	—	+	+	+	+	+	?	?
Flustra foliacea	+	+	+	+	—	—	—	—	—	+	+	+	+	—

B

Appendix II (cont).

Species	J	F	M	A	M	J	J	A	S	O	N	D	Distribution Nor.	Distribution Med.
Micropora coriacea	+	+	+	+	+	+	+	+	+	+	+	+	–	+
Cellaria fistulosa	+	+	+	+	–	–	–	–	+	+	+	+	+	+
Cellaria sinuosa	+	+	[illegible]	+	+	+	–	+	+	+	+	+	?	?
~~Scrupocellaria~~ scruposa	–	–	–	–	–	–	+	+	+	+	+	+	+	+
Scrupocellaria reptans	+	+	+	–	+	+	+	+	+	–	+	+	+	+
Bicellariella ciliata	–	–	+	+	+	+	+	+	+	+	+	–	+	+
Bugula avicularia	–	–	–	–	–	+	+	+	+	+	+	–	–	+
Bugula plumosa	–	–	–	–	–	–	–	–	+	+	–	+	+	+
Bugula flabellata	+	–	–	–	+	+	+	+	+	+	–	–	+	+
Bugula turbinata	–	–	–	–	–	–	–	–	+	+	+	–	?	?
Membraniporella nitida	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Cribrilina punctata	+	+	+	+	+	+	–	+	+	[illegible]	+	+	+	+
Cribrilina annulata	–	–	–	–	–	–	+	–	–	–	+	–	?	?
Cribrilaria radiata	+	+	+	+	+	+	+	+	+	+	+	+	–	+
Figularia figularis	+	–	+	+	[illegible]	+	–	+	+	+	+	+	–	?
Hippothoa distans	+	+	+	–	+	+	–	–	+	+	[illegible]	+	+	+
Hippothoa hyalina	+	+	+	+	+	+	++	+	+	+	+	+	+	+
Chorizopora brongniarti	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Haplopoma graniferum	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Schizotheca fissa	–	+	–	–	–	+	+	+	+	+	–	+	–	+
Reptadeonella violacea	+	–	+	+	+	+	–	–	+	–	–	–	+	+
Escharella immersa	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Escharella ventricosa	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Escharella variolosa	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Schizoporella unicornis	+	–	–	–	–	+	+	+	+	+	+	+	+	+
Schizoporella discoidea	+	–	–	–	–	–	–	–	+	+	+	–		
Schizomavella auriculata	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Schizomavella linearis	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Escharina spiniferum	+	+	+	+	+	+	+	+	–	–	–	–	?	?

C

Appendix II (cont.)

Species	J	F	M	A	M	J	J	A	S	O	N	D	Nor.	Med.
Escharina vulgaris	−	−	+	−	+	−	−	+	−	−	−	+	−	+
Escharina johnstoni	+	−	−	−	+	−	−	−	−	−	−	−	?	?
Herentia hyndmanni	−	−	−	−	−	−	−	−	−	−	+	−	−	+
[illegible]orina pertusa	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Hipporporidra edax	−	−	+	−	−	−	−	−	−	−	−	−	?	?
Lepralia foliacea	−	−	−	−	−	−	+	−	+	−	−	−	+	+
Microporella ciliata	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Fenestrulina malusi	+	+	+	+	[illegible]	+	+	+	+	+	+	+	+	+
Palmicellaria skenei	−	−	−	+	−	−	−	−	+	−	−	+	+	+
Porella concinna	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Porella minuta	−	+	−	−	−	−	−	−	−	−	−	+	−	+
Smittina landsborovi	+	−	+	−	−	−	−	−	−	+	+	−	+	+
Smittina cheilostomata	+	−	+	−	−	+	+	+	+	+	+	+	−	+
Parasmittina trispinosa	+	+	+	+	+	+	[illegible]	+	+	+	+	+	+	+
Smittoidea reticulata	+	+	−	−	−	+	−	+	+	−	+	+	++	+
Phylactella collaris	−	−	−	−	−	+	−	+	+	−	+	+	?	?
Phylactella labrosa	−	−	−	−	−	−	−	+	−	−	−	−	?	?
Escharoides coccineus	+	+	+	+	[illegible]	+	+	+	+	+	+	−	+	+
Umbonula littoralis	−	+	−	−	−	−	−	−	+	+	+	−	+	−
Celleporaria dichotoma	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Celleporaria pumicosa	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Osthimosia avicularis	+	+	+	+	+	+	+	+	+	+	+	−	+	+
Cellepora costazii	+	+	+	+	+	+	+	+	+	+	+	+	+	+
Alcyonidium hirsutum	+	+	+	+	−	−	−	−	+	+	+	+	+	−
Alcyonidium gelatinosum	+	+	+	−	−	−	−	−	−	−	+	+	+	−
Alcyonidium variegatum	+	+	+	+	+	−	−	+	+	+	+	+	?	?
Alcyonidium polyoum	+	+	−	−	−	−	−	+	−	−	−	−	?	?
Alcyonidium mytili	+	+	+	+	+	+	−	−	−	+	+	+	?	?

D

Appendix II (cont.).

Species	J	F	M	A	M	J	J	A	S	O	N	D	Nor.	Med.
Flustrellidra hispida	-	+	+	+	+	+	+	-	-	-	-	-	+	+
Bowerbankia pustulosa	-	-	-	-	-	-	-	-	+	-	-	-	-	+
Bowerbankia imbricata	-	-	-	-	-	+	+	+	+	-	-	-	+	+
Bowerbankia gracilis	-	-	-	-	-	+	-	-	-	+	-	-	+	+
Buskia nitens	-	-	-	-	-	-	-	+	+	+	+	-	+	+
Valkeria uva	-	-	-	-	-	-	-	+	+	-	-	-	+	+
Triticella koreni	-	-	+	-	-	-	-	+	-	+	+	-	+	-
Hypophorella expansa	-	-	-	-	-	-	-	-	-	+	-	-	+	+
Nolella dilatata	+	-	-	-	-	-	-	-	-	-	+	+	+	+

Appendix III. Embryo Colour in Manx Ectoprocta.

Cyclostomata: Ovicells containing early embryos may appear yellowish (particularily in the Crisiidae) but fully developed embryos are usually colourless.

Cheilostomata:

Species	Colour of embryos: Munsell notation	Colour of embryos: English description.
Aetea anguinea	-	golden-yellow
Aetea sica	-	golden-yellow
Eucratea loricata	-	white
Alderina imbellis	10·0 YR $\frac{8\cdot5}{6}$	yellowish-white
Callopora lineata	5·0 R $\frac{5}{10}$	red
Callopora dumerili	2·5 YR $\frac{6-7}{14}$	orange
Callopora aurita	-	white
Callopora craticula	2·5YR $\frac{6}{12}$, 7·5 R $\frac{6}{10}$, 10·0 R $\frac{6}{8}$	red
Amphiblestrum flemingi	5·0 - 10·0 YR $\frac{7}{6-10}$	orange
Cauloramphus spiniferum	early 2·5 YR $\frac{6-7}{12-14}$; late 7·5 -10·0 YR $\frac{7}{6}$	orange
Flustra foliacea	7·5 YR $\frac{8}{6}$	orange
Hincksina flustroides	-	yellow-orange
Micropora coriacea	2·5 YR $\frac{6}{12}$	orange
Cellaria sinuosa	2·5 Y $\frac{8}{4}$	golden-yellow to pale yellow
Cellaria fistulosa	2·5 Y $\frac{8}{4}$	" " " "
Scrupocellaria scruposa	-	red
Scrupocellaria reptans	-	deep red
Bicellariella ciliata	-	white
Bugula avicularia	-	yellow
Bugula plumosa	-	yellow
Bugula flabellata	-	bright golden-yellow
Bugula turbinata	-	" " "
Membraniporella nitida	2·5 - 7·5 YR $\frac{6-7}{8-10}$	yellow-orange
Cribrilina punctata	10·0 R- 2·5 YR $\frac{5-7}{8-10}$	red
Cribrilina annulata	5·0 YR $\frac{6}{10}$	orange
Cribrilaria radiata	-	dull orange-red

Appendix III . (cont.)

Species	Colour of Embryos Munsell notation	English description
Figularia figularis	embryos 10·0 R– 2·5 YR 6/10-14 larvae 5·0 YR–, 7·5 YR 6/8	red-brown
Hippothoa distans	–	yellow
Hippothoa hyalina	2·5 – 5·0 Y 8-9/3-8	yellow
Chorizopora brongniarti	9·0 R 5/10	red
Haplopoma graniferum	2·5 – 5·0 R 7/4	pink
Schizotheca fissa	–	red
Rentadeonella violacea	7·5 – 10·0 YR 5/10	brown
Escharella immersa	2·5 R 5/6, 5·0 R 7/6, 7·5–8·0 R 5/12, 10·0 R 7/4 .	red
Escharella ventricosa	–	white
Escharella variolosa	2·5 YR 6-7/10, 10·0 YR 7/10	orange-yellow
Schizoporella unicornis	–	dark orange-red
Schizoporella discoidea	–	red
Schizomavella auriculata	2·5 YR 6/14 –10·0 YR 6/10	brownish-red
Schizomavella linearis	2·5 YR 6/12, 2·5 YR 6/8, 5·0 YR 6/8, 7·5 R 6/10, 10 0 R 6/10	red, orange-red
Escharina spiniferum	2 5 YR 7/10; 5·0 YR 7/10; 10·0 R 6/10	orange-red
Escharina vulgaris	–	golden-yellow
Escharina hyndmanni	–	pale red
Hippoporina pertusa	1·0 YR 6/14; 2·5 YR 6/10, 5 0 YR 7/10	bright orange
Hippoporidra edax	3 0– 5 0 YR 7/10	orange-yellow
Lepralia foliacea	–	bright yellow
Microporella ciliata	early 10·0 R 6/10, 5/8, 5·0 YR 7/10 late 2 5 YR 6/10, 7·5–10·0 YR 5-10/8-10	red
Fenestrulina malusi	early 2·5 YR 5/10 late 10·0 YR 6-7/8-10	golden-yellow
Palmicellaria skenei	7·5 YR 6/8	orange
Porella concinna	2·0 YR 5/10 7·5 R 5/12 10·0 R 6/14	red
Porella minuta	–	brownish-yellow
Smittoidea reticulata	–	orange red
Smittina landsborovi	–	red
Smittina cheilostomata	–	red
Parasmittina trispinosa	early ca. 7·5 R 5/14 late 5·0 YR 7/10	bright red yellow-orange
Phyllactella labrosa	–	orange-red
Phyllactella collaris	–	yellow

Appendix III (cont)

Species		Colour of Embryos: Munsell notation	Colour of Embryos: English description
Escharoides coccineus	early	3·0 R 3/8	
	late	5·0 YR 4/10	maroon
Umbonula littoralis		–	red
Cellepora dichotoma		2·5 YR 6/10	orange-red
Cellepora pumicosa		10·0 R 4/– 2·5 YR 5/10	red
Onthimosia avicularis		10·0 YR 7-8/9-10	yellow
Celleporina costazii	early	7·5 R 3/10	
	late	10·0 R 5/10	red
Ctenostomata:			
Alcyonidium hirsutum		–	white
Alcyonidium gelatinosum		–	white
Alcyonidium variegatum		–	orange-red
Alcyonidium polyoum		–	white
Alcyonidium mytili		10 0 RP 8/2	pale pink
Flustrellidra hispida		–	white
Bowerbankia pustulosa		–	yellow
Bowerbankia imbricata		–	yellow
Bowerbankia gracilis		–	pink
Buskia nitens		–	yellow
Valkeria uva		–	colourless
Nolella dilatata		–	white

Appendix IV. Key to the Ectoprocta of the Isle of Man.

This key should be used in conjunction with the text and figures of Hincks (1880) "British Marine Polyzoa", or Marcus (1940) "Danmarks Fauna:Mosdyr".

1	Colony not calcareous or strongly chitinised	2
	Colony calcareous or strongly chitinized	18
2	Colony often fleshy, zooids contiguous except, perhaps, at colony edge. Aperture frontal.	3
	Zooids in more or less branching chains or with separate zooecia united by filiform stolons.	4
3	Aperture with cuticular spines	*Flustrellidra hispida*
	Aperture without spines	Family Alcyonidiidae (p XIX)
4	Colony a network formed of anastomosing prolongations from the autozooecia which occupy the nodes of the net.	5
	Colony a branched stolonial axis of long kenozooids placed end to end and either bearing the autozooecia directly or on small lateral kenozooids	8
5	Peristome short	6
	Peristome long (over 500μ)	7
6	Autozooid with fringe of filamentous prolongations	*Arachnidium fibrosum*
	Autozooid without prolongations	*A. hippothooides.*
7	Peristome about 550μ, 10 tentacles	*Nolella pusilla*
	Peristome from 550-1450μ, 16-20 tentacles	*N. dilatata*
8	Boring	9
	Not boring	10
9	Boring in membranous worm tubes	*Hypophorella expansa*
	Boring in calcareous materials	*Penetrantia concharum*

10 Stolonial kenozooids rarely of uniform thickness usually filiform and slightly swollen at distal end. The distal end gives rise either to new, lateral stolons (usually two) or to lateral series of small kenozooids which bear the autozooids. Autozooids often mobile. No gizzard. 11

Stolons generally robust and of uniform thickness. Autozooids are budded directly from the stolon either singly, or in one, two or three more or less helicoidal series. Autozooids immobile. Gizzard present. 14

11 Autozooids deciduous, aperture quadrangular, no membranous area or bilateral symmetry 12

More or less bilaterally symmetrical and with membranous area 13

12 Autozooid tapering towards attachment. Autozooid mobile. <u>Walkeria tremula</u>

Autozooid narrows abruptly at attachment. Autozooid not mobile <u>Walkeria uva</u>

13A Autozooids on a long, rigid peduncle usually at least half the length of the autozooid. Stolons incrusting. <u>Triticella koreni</u>

Autozooids not on long peduncle. Colony erect. <u>Mimosella gracilis</u>

14 Autozooid with an anterior membranous area and attached directly to the stolon. Autozooids not cylindrical or in regular series. <u>Buskia nitens</u>

Autozooids without membranous area, cylindrical and contracted at their bases which are attached directly to the stolon. 15

15 Autozooids closely contiguous, tubular, in two parallel helicoidal series divided into close groups. Erect <u>Amathia lendigera</u>

Autozooids not closely contiguous, budded sparsely or in small groups which ifhalicoidal are not as regular as <u>A. lendigera</u>. Colony erect or rampant. 16

16 Erect <u>Bowerbankia pustulosa</u>

Rampant or encrusting 17

17 Autozooid with tail-like prolongation near its attachment to the stolon. 8 tentacles <u>Bowerbankia gracilis</u>

Autozooid without "tail". 10 tentacles <u>Bowerbankia imbricata</u>

(Colony calcareous or strongly chitinized.)

18 Erect 19

Encrusting 42

19 With articulated, chitinous joints 20

Unjointed 23

20 Branchs of one or two series of zooids. Aviculariae absent. Embryos develop in ovicells. Orifice terminal
Fam. Crisiidae.(pXIII)

Branchs tubular with numerous zooids in regular series. Aviculariae and ooecia may be present. Orifice frontal 21

21 Orifice (aperture) with broad tooth in lower lip <u>Cellaria sinuosa</u>

Orifice lower lip slightly incurved but without broad tooth. 22

22 Zooecia contiguous. <u>Cellaria fistulosa</u>

Zooecia distant ,seperated by a raised line. <u>Cellaria salicornioides</u>

23 Colony a single series of zooecia 24

Colony of several series of zooecia 27

24 Zooecia seperated by "stolons", spines present around aperture.
<u>Beania mirabile</u>

Zooecia not seperated by "stolons", no spines 25

25 Branchs lateral, large membranous frontal area occupying most of the front of zooid. <u>Pyripora catenularia</u>

Branchs arise from front of zooid below aperture. No membranous frontal area 26

26 Colony spreading over support by incrusting stolons.
<u>Scruparia chelata</u>

Colony spreading over support by attaching, incrusting zooids.
<u>Scruparia ambigua</u>

27 Colony stout, inflexible 28

Colony plant-like, flexible. 35

28 Colony a series of erect, anastomosing, foliaceous, bi-lamellar plates
" <u>Lepralia</u>" <u>foliacea</u>

Colony not as above 29

29 Aviculariae and/or ooecia present. Zooecia not tubular. Aperture variable but not on an erect tube. No common calcareous matrix. 30

Aviculariae and ooecia never present, ovicells may be present. Zooecia tubular, embedded in a common calcareous matrix. Aperture circular on an erect tube. 33

30 Branchs flat zooecia regular. Palmicellaria skenei

Branchs rounded or oval not flat. 31

31 Spatulate aviculariae present 32

Spatulate aviculariae absent Porella compressa

32 Ooecium punctate. Rostrum short, stumpy with an avicularium to one side. Small circular aviculariae present on colony Cellenoraria dichotoma

Ooecium entire. Rostrum well-developed, pointed, with central avicularium. No circular aviculariae. Omalosecosa ramulosa

33 Zooecia seperate, on all sides of branch. 34

Zooecia contiguous, basal side of branch without zooecia Tubulipora sp (p. XIV)

34 Colony mushroom shaped. Ovicell terminal Tubulipora penicillata

Colony not as above. Ovicell lateral. Entalophora clavata

35 Zooecia in two layers (back to back) 36

Colony a single series of zooecia 26

Colony of several series of zooecia all on the same side 38

36 Zooecia in a single series of paired (back to back) Eucratea loricata

Zooecia in several series in each branch 37

37 Zooecia with spines Flustra foliacea

Without spines Securiflustra securifrons

38 Aviculariae pedunculate 41

Aviculariae sessile 39

39 Zooecia without scutum — *Scrupocellaria scruposa*

Zooecia with scutum — 40

40 Scutum entire, rounded — *Scrupocellaria scrupea*

Scutum branched — *Scrupocellaria reptans*

41 Less than 6 spines on each zooecium, if biserial less than 5 — *Bugula* spp. (p. XVIII)

More than 6 spines on each zooecium, buserial — *Bicellariella ciliata*

(Colony calcareous , encrusting)

42 Aperture terminal, no ooecia(calcified), no aviculariae or vibraculae. Autozooecia tubular. — 43

Aperture not terminal, autozooecia not tubular, ooecia (calcified) , aviculariae or vibraculae may be present — 61

43 Zooecia seperate, no common calcareous matrix. — 44

Zooecia not seperated , in common calcareous matrix — 46

44 Erect part of zooecium with very fine annulations — 45

Without such annulations — *Aetea truncata*

45 Zooecia may be curved, top of erect part swollen and spoon-like. — *Aetea anguinea*

Zooecia straight, and not spoon-like at tip — *Aetea sica*

46. Colony wart-like or mammiform. — 47

Colony not as above — 50

47 Colony mammiform, sometimes compound. Top of mammiform disk rounded and furrowed. Zooecia in multiserial rows. Colony appears to be of superimposed layers — *Domopora truncata*

Colony may be sub-conical but not mammiform, zooecia uniserial, colony not ridged or furrowed — 48

48 Zooecia connate in radiating rows — *Lichenopora radiata*

Zooecia not connate — 49

49 Zooecia frequently with two or more spines. Alveoli thick walled. Ooeciostome little raised and not trumpet-like. — *Lichenopora hispida*

Zooecia rarely more than one spine. Alveoli thin walled. Ooeciostome trumpet-shaped wtih a broad flange. — *Lichenopora verrucaria*

50 [illegible]

50 Colony entire, circular or lobulated. 51

Colony branched, linear, not entire 53

51 Dwarf zooids present *Diplosolen obelia*

Dwarf zooids absent 52

52 Colony with a thin calcareous lamina around edge. Some autozooids with aperture closed by a calcareous plug. *Diastopora patina*

Colony without calcareous lamina zooecia reaching to colony edge. All zooecia apertures open. *Diastopora suborbicularis*

53 Colony retiform, composed of numerous anastomosing branches seperated by elongate spaces, branches frequently giving rise to short erect processes terminating in a cellular apex. *Proboscina incrassata*

Colony not retiform and without erect cellular processes. 54

54 Zooecia in single series for much of the colony. *Stomatopora granulata*

Zooecia in more thana single series for most of the colony 55

55 Zooecia in not more than two rows except near the ovicell 56

Zooecia in more than two rows for most of the colony 57

56 Dichotomously branched. Apertures open upwards *Diaperoecia johnstoni*

Rarely branched. Apertures open laterally *Stomatopora incurvata*

57 Ooeciostome associated with a normal autozooid. Many rows of zooecia, frequently in transverse series. Family Tubuliporidae (p. XV)

Ooeciostome independent of other zooecia 58

58 Zooecia very large in 1-4 series, sometimes in transverse rows *Diaperoecia major*

Zooecia not unusually large, in more than 4 series, not usually in transverse rows 59

59 Ooeciostome veryshort *Tubulipora aperta*

Ooeciostome lacking 60

60 Branchs radiating from a com on point, zooecia short and stout. Purple when fresh. *Tubulipora lobulata*

Branches serpentine, zooecia slender. White in colour *Oncousoecia dilatans*

61 Zooecia seperate, joined by elongate "stolons". 62

Zooecia contiguous 64

62 Zooecia erect, with spines *Beania mirabile*

Zooecia encrusting without spines 63

63 Aperture arched above and with notch in lower margin. More or less well-developed keel on front of zooecium *Hippothoa divaricata*

Aperture sub-ovate, broader above than below, Keel Absent *Hippothoa distans*

64 Front wall of zooecia not completely calcified leaving a membranous area. 65

Front wall of zooecia fully calcified but may be punctate 67

65 Avicularium mandible pointed, aviculariae sometimes absent. Family Membraniporidae (p.XV)

Avicularium mandible rounded, aviculariae usually present. 66

66 Autozooid with 2 - 4 spines *Flustra foliacea*

12 - 14 spines *Hincksina flustroides*

67 A pair of vibraculae or aviculariae, (one in a similar position on each side of the autozooid) on most zooids, other aviculariae may be present. 68

Aviculariae or vibraculae, when present, rarely paired, or if paired dissimilar in size and appearance 77

68 Vibraculae or vibraculoid aviculariae present 69

Aviculariae not vibraculoid. 72

69 Frontal calcified area punctate, lower margin of aperture straight 70

Frontal surface entire, lower margin of aperture with sinus (notch) 71

70. 5-6 marginal spines. Avicularia normally present. Ooecia globose, keeled. _Cribrilaria radiata_

5 marginal spines. Avicularia absent. Ooecia punctate. _Puellina gattyae_

71 Vibraculum on each side of orifice _Mastigophora dutertrei_

Vibraculoid avicularium well below orifice _Escharina vulgaris_

72 Colony an irregular mass of zooecia 73

Colony plate-like, zooecia regular 74

73 Stout rostrum bearing avicularium present in many zooecia _Osthimosia avicularis_

No rostrum below the aperture. _Celleporina costazii_

74 Frontal surface of zooecium punctate _Cribrilina punctata_

Frontal surface entire 75

75 Marginal spines presentaround aperture 76

No spines _Schizoporella unicornis_

76 Large avicularia, lower margin of aperture with at least two indentations _Escharoides coccineus_

Avicularia small, inconspicuous, lower margin of aperture with one notch. _Schizomavella linearis_

77 (Frontal calcareous, avicularia or vibracula, if present not paired)

An avicularium present directly below aperture 78

Avicularium if present not directly below aperture 86

78 Avicularium on a pointed rostrum below apeture 79

Avicularium not on pointed rostrum. 80

79 Spatulate avicularia scattered amongst zooecia _Osthimosia armata_

Spatulate avicularia absent _Celleporaria pumicosa_

80 Avicularian mandible pointed 81

Avicularium mandible rounded or spatulate 83

81 Avicularium erect, on side of pointed mucro, aperture with sinus(notch) which does not have tooth on inner margin. _Schizomavella linearis_ var. _hastata_

Avicularium not as above, aperture with or without sinus, if sinus present a prominent tooth present on its inner margin. 82

82 Aperture without sinus, avicularium mandible directed towards aperture.Ooecia never present. Reptadeonella violacea

Aperture with sinus, avicularium mandible directed away from aperture.Ooecia may be present. Smittoidea reticulata

83 Well-developed collar around aperture 84

Collar absent 86

84 Aperture with sinus with median denticle, a small rounded avicularium partly enclosed in sinus Smittina landsborovi

Without sinus and denticle. 85

85 Ooecia never present. Zooecia in lines, mainly littoral Cryptosula pallasiana

Ooecia sometimes present. Zooecia quincuncial. Sub-littoral "Lepralia" foliacea

86 Sinus but no denticle in aperture. 2-4 spines. Avicularium immediately below aperture lower lip. Schizomavella auriculata

Sinus absent, Never more than 2 spines, avicularium on lower margin of aperture 87

87 Zooecia large. Avicularium on a broad tooth. Mucro never present Porella concinna

Zooecia very small. Avicularium on rounded eminence. Mucro sometimes present. Porella minuta

88 Aviculariae or vibraculae present 89

Aviculariae and vibraculae absent 101

89 Colony an irregular mass of zooecia, aperture key-hole shaped Hippoporidra edax

Colony regular, zooecia ordered, aperture not key-hole shaped. 90

90 Frontal surface of zooid covered by system of fused spines seperated by slits. Membraniporella nitida

Frontal punctate or entire. 91

91 Movable spine below aperture Escharina spiniferum

Spines, if any, only around the aperture 92

92 Semi-lunate pore below aperture Microporella ciliata

No such pore 93

93 Vibraculum with long "flagellum" on each zooecia
Matigophora hyndmanni s

No true vibraculum present although avicularia may have elongate mandible 94

94 Marginal spines 95

No marginal spines 96

95 Avicularia on distinct area separate from zooecium. Ooecia with wedge-shaped frontal fissure. 6 spines.
Schizotheca fissa

Avicularia lateral on zooecia surface, No fissure in ooecium. 6-7 spines Schizoporella discoidea

96 Very large spatulate avicularia scattered among zooecia 97

Large spatulate avicularia absent 98

97. Frontal surface with rows of tiny punctures. No rostrum.
Figularia figularis

Frontal surface entire. Rostrum bearing small avicularium to one side below aperture Osthimosia armata

98 Avicularia with pointed mandible either on ooecia or on flat areas above the zooecia Chorhizopora brongniarti

Avicularia on frontal surface of zooecia 99

99 Aperture without sinus Hippoporina pertusa

Aperture with sinus 100

100 Aperture with broad flat collar Escharina simplex

No such collar Escharina alderi

101 Marginal spines 102

No marginal spines 109

102 Frontal surface punctate, punctures in rows, Cribrilina annulata

Frontal without rows of punctures 103

103 Semi-lunate pore below aperture Fenestrulina malusi

No such pore 104

104 Aperture with median denticle on lower lip, no sinus 105

Aperture with sinus, no denticle 108

105 Denticle bifid 106

Denticle not bifid 107

106 Zooecia strongly convex. 4 spines *Escharella ventricosa*

Zooecia almost flat. 6-8 spines *Escharella immersa*

107 Orifice slightly broader than high, lower side only slightly raised *Escharella variolosa*

Lower lip of orifice raised into a screen-like plate. Orifice plainly broader than high *Escharella abyssicola*

108 Zooid without frontal spine. Ooecia with fissure, collar well-developed around aperture. *Schizotheca fissa*

Articulated spine on front of zooecia. Ooecia globose, entire Collar not well developed. *Escharina spiniferum*

109 Circular pore present below aperture 110

No such pore 111

110 Frontal wall with few pores, scattered around margin *Haplopoma impressum*

Frontal with many pores covering it wholly except in the region of the umbo *Haplopoma graniferum*

111 Walls hyaline, thin. Ooecia on dwarf zooecia *Hippothoa hyalina*

Walls not as above nor are ooecia on dwarf zooecia 112

112 Orificial collar absent or poorly developed 113

Collar well developed 117

113 Aperture semi-circular, lower margin straight, usually with a knob on each side of the aperture. *Microporella coriacea* Frontal punctate in all zooecia

Aperture as above, only young zooecia with frontal punctate, in older zooecia frontal granular or ridged. *Lepralia adpressa*

Aperture not as above 114

114 Aperture without sinus or median denticle 116

With sinus or without sinus, with median denticle 115

115
Without sinus — Escharella variolosa
With sinus — Smittina Cheilostomata

116 Aperture almost circular, ooecia may be present
No radiating ridges on frontal surface — Hipporina pertusa

Lower lip incurved. Ooecia never present, frontal surface with radiating ridges — Umbonula littoralis

117 Collar around aperture flat — Escharina simplex

Collar around aperture raised — 118

118 Aperture with sinus — 119

Aperture without sinus — 120

119 Median denticle in lower lip of aperture — Smittina cheilostomata

No median denticle — Hippothoa hyalina

120 Aperture key-hole shaped — Cryptosula pallasiana

Not as above — 121

121 Aperture with median denticle — Phylactella labrosa

Not as above — 122

122 Zooecia in common calcareous crust Zooecia wall smooth except for the smooth erect part bearing the aperture. Ooecia smooth — Lagenipora socialis

Zooecia in radiating lines, no common crust. Wall smooth, aperture not on erect part of zooecium. Ooecia punctate — Phyllactella collaris

Key to the Crisiidae recorded from the Isle of Man

1.	Most or all the internodes of a single zooid	2
	Internodes of a single zooid only in the oldest parts of the colony	3
2	Spines present	*Crisidia cornuta*
	No spines	*Filicrisia geniculata*
3	Branches, particularily in youngest parts of the colony, strongly incurved.	*Crisia eburnea*
	Branches not, or very slightly, incurved	4
4	With spines	*Crisia aculeata*
	Without spines	5
5	Joints jet-black. Ooeciostome a very short tube	*Crisia denticulata*
	Joints colourless. Ooeciostome long, funnel-shaped tube.	*Crisia ramosa*.

Key to the Tubuliporidae recorded from the Isle of Man.

1. Colony with neat, transverse rows of connate zooecia 2

Colony without, or with only short transverse rows of zooecia 4

2 Oooeciostome not recumbent on a zooecium, funnel shaped often with one side folded over, much larger than a zooid and opening upwards or obliquely horizontal.
<u>Tubulipora plumosa</u>

Oooeciostome tube recumbent on a zooecium. Ooeciopore opening horizontal or downwards 3

3 Ooeciostome mouth larger than a zooecial aperture, and opens horizontally <u>Tubulipora liliacea</u>

Ooeciopore concealed, smaller than an apertures and facing down. <u>Tubulipora phalangea</u>

4 Short encrusting part of colony gives rise to erect mush-room-like branches <u>Tubulipora pennicilliata</u>

No erect branches 5

5 Colony broad, lobed or pear-shaped. If lobed, lobes fan-like. Ooeciostome tube wide or opens upwards.
<u>Tubulipora aperta</u>

Colony broad, narrowest part has 6 rows of zooecia which increases to 10 rows near an ovicell. Ooeciostome without tube <u>Tubulipora lobulata</u>

Key to the Ectoprocts of the Family Membraniporiidae recorded from the Isle of Man. (modified from Marcus 1940)

1. Aviculariae and ooecia absent 2

Aviculariae or ooecia or both present 9

2 Zooecia rectangular, Frontal area four cornered *Membranipora membranacea*

Zooecia not regularily rectangular, frontal area rounded 3

3 Calcareous part of zooecia with pore-like spots 4

Not as above 5

4 'Spots' over whole of calcareous part of zooecium *Electra pilosa*

Few 'spots' loosely scattered *Electra hastingsae*

5 Operculum white, calcareous *Electra crustulenta*

Operculum not calcareous 6

6 Zooecia taper markedly below mid-line 7

Zooecia not tapering downwards 8

7 Frontal area with spines *Electra hastingsae*

No spines *Pyripora catenularia*

8 Cryptocyst slightly developed around edge of frontal area which is smooth or beaded and may have fine spines. Among zooecia are small calcareous mounds with central triangular or rounded hollows *Conopeum reticulum*

Thin calcareous cryptocyst developed below membranous frontal area particularily at its basal endn No mounds or hollows among the zooecia but occasionally node-like spines occur at corners of zooecia. *Acanthodesia tenuis*

9 Crypocyst absent or poorly developed never covering more thana third of the frontal area, 10

Strongly developed cryptocyst never covers less than a third of the frontal area. 13

10 Avicularia and spines aabsent 11

Aviculariae and spines both present 12

11 4 Spines *Alderina solidula*

No spines *Alderina imbellis*

12 Most ooecia with avicularium obliquely on upper surface
Tegella unicornis

No avicularia on upper surface of ooecia 13

13 Pedunculate aviculariae. *Caulorampnus spiniferum*
Not as above 14

14 Avicularium absent, more than 20 delicate spines
Callopora discreta

Avicularia present, less than 20 spines. 15

15 Ooecia front wall with curved or angular ridge not glistening. Embryo colour not orange-yellow 16

Ooecial wall smooth, glistening. Embryos orange-yellow
Callopora dumerili

16 At least 6 spines. With/or without a single avicularium above the ooecium. Embryos red. 17

4 spines. Often two aviculariae above each ooecium, embryos white *Callopora aurita*

17 6-12 spines. Spines not flat, glistening or cage-like across front of zooecium. *Callopora lineata*

12-14 flat, glistening spines form a cage-like arch over front of zooecium *Callopora craticula*

18 Aviculariae absent 19

Aviculariae present 21

19 4-6 spines on top of zooid and sometimes a chitinous vibraculoids spine on front wall *Megapora ringens*

No spines 20

20 Zooid margin slightly raised, granular. Large sub-triangular noc nodule at base of each zooid. *Ammatophora nodulosa*

Margin of zooid raised and beaded, no sub-triangular nodules *Rosseliana rosseli*

21 Avicularium mandible tri-angular, pointed. Spines simple never branched. 22

Avicularian mandible rounded, spatulate, lowest pair of spines branched. *Larnacicus corniger*

22 6-8 spines, one spine often stouter and longer than others, Often 2 avicularia per zooecium *Amphiblestrum flemingi*

Spines sometimes absent, never one more developed than the others. Only 1 avicularium per zooecium 23

23 Spines present. Avicularium on frontal area margin, mandible long, tapering to fine point, *Ramphonotus minax*

No spines, avicularise between zooecia, mandible acute *Amphiblestrum solidum*

Key tô the species of Bugula recorded from Manx waters.

1	Biserial	2
	Multiserial	3
2	2 spines on outer distal angle of zooecia. 1 on inner angle. Avicularium $\frac{1}{3}$-$\frac{1}{2}$ way down opesium is longer than the zooecial width. Beak elongated, downcurved	Bugula avicularia
	1 spine on each distal angle. Avicularid attached immediately below spine on outer wall. Beak sharply downcurved.	Bugula turbinata
	1, more or less marked, unjointed spine on the outer distal angle, inner angle unarmed. Avicularium $\frac{1}{3}$-$\frac{1}{2}$ way down opesium. Beak slightly downcurved.	Bugula plumosa
3	One spine on each distal angle	Bugula turbinata
	3 spines on outer distal angle ,2 on others	4
4	Avicularium beak down-curved. Avicularium length equals or surpasses width of two zooecia	Bugula calathus
	Avicularium beak abruptly down curved. No avicularium equals 2 zooecia widths.	Bugula flabellata

Key to the Alcyonidiidae recorded from Manx waters.
(modified from Bobin and Prenant 1956)

1 Colony incrusting, terraceous, zooecia with numerous filiform imperforate papillae around the aperture and peristomial region. *Alcyonidium parasiticum*

Colony incrusting or erect, not terraceous in appearance or consistency. 2

2 Each zooid has 5-6 conical, imperforate papillae around the aperture. *Alcyonidium hirsutum*

Without such papillae 3

3 Orifice on long peristomial tube. 4

Peristomial tube very short or absent 5

4 Peristomial tube $\frac{1}{5} - \frac{3}{4}$ zooid length when contracted. Orifice circular. Colony incrusting, generally entire but may give off series of zooecia near the edge of the colony *Alcyonidium mammillatum*

Peristomial tubes long, but not as long as in the previous species. Zooecia in disjointed linear series at the edge of the colony but are irregularily placed towards the centre. Orifice often tri-foliate when contracted. *Alcyonidium albidum*

5 Colony erect, *Alcyonidium gelatinosum*

Encrusting 6

6 Colony of uniform thickness to edge of colony, zooecia perpendicular to support. Embryos bright orange *Alcyonidium variegatum*

Colony thinner towards edge where zooecia are parallel to support. Embryos white or pale pink 7

7 Embryos white. Zooecial walls often difficult to see in older parts of colony. 17-18 tentacles *Alcyonidium polyoum*

Embryos pink. Old zooecia walls whitish and obvious to naked eye. 19-20 tentacles. *Alcyonidium mytili*