DISTRIBUTION AND ABUNDANCE OF LARVAL *COQUILLETTIDIA PERTURBANS* IN A FLORIDA FRESHWATER MARSH[1]

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ABSTRACT. All instars of larval *Coquillettidia perturbans* were found in the same habitats, but early instar larvae were more aggregated than later instars. Larvae were most numerous in areas dominated by arrow-arum (*Peltandra virginica*) and maidencane (*Panicum hemitomon*), less so in areas dominated by sedges (*Carex* spp.) and miscellaneous mixed vegetation, and least abundant in pickerelweed (*Pontederia cordata*) areas. Larvae were uncommon in open water or in areas dominated by small floating plants such as water fern (*Salvinia rotundifolia*), duckweed (*Lemna minor*) and mosquito fern (*Azolla caroliniana*). Larval concentrations were greatest in water 35–70 cm deep. There was also a tendency for them to concentrate in areas beyond 25 m from shore. Larvae were log-normally distributed in favorable sites and became progressively more aggregated as sites became less favorable.

INTRODUCTION

The widely ranging *Coquillettidia perturbans* (Walker) is an important pest near its permanent freshwater marsh breeding sites, and it is intimately associated with epidemic and epiornithic foci of Eastern equine encephalomyelitis virus in the eastern United States (Morris 1988). The larvae of *Cq. perturbans* attach to the submerged roots of emergent aquatic vegetation. Larvae occur primarily in the benthic zone of well established marshes and swamps which characteristically have low dissolved oxygen, low pH and a substantial organic detrital bottom cover (McNeel 1932, Brower 1953, Callahan and Morris 1987a). They are typically associated with cattails (*Typha* spp.) and sedges (*Carex* spp.), and in temperate climates are often abundant on floating mats (Allan et al. 1981, Batzer and Sjogren 1986, Olds et al. 1989).

In Florida, floating cattail mats produce more *Cq. perturbans* than rooted cattails (Slaff and Haefner 1985) but the species is also a prolific breeder in many other southern plant communities (Bidlingmayer 1968, Lounibos and Escher 1983, Slaff and Haefner 1985).

It is difficult and expensive to larvicide effectively in *Cq. perturbans* habitats. Nonetheless, the Metropolitan Mosquito Control District of St. Paul, Minnesota, controls larval *Cq. perturbans* with methoprene briquettes placed at the edges of cattail mats and in holes within the mats (R. D. Sjogren, personal communication). This treatment is economical because larvae in these mats occur only within 1 m of the cattail edges or openings (Batzer and Sjogren 1986). Larval distributions in other than floating cattail mats in Minnesota have not been examined. If larvae in Florida aggregate as they do in Minnesota, then perhaps briquettes or other control methods could be economically applied in areas of high larval concentration.

Thus, studies were begun to: 1) describe the spatial distributions of all larval instars of *Cq. perturbans*, and 2) determine the factors which influence these distributions.

MATERIALS AND METHODS

The 4.5-ha study site was part of an extensive freshwater marsh system covering approximately 10,000 ha northeast of the city of Lake Alfred, FL. The site was dominated by maidencane grass (*Panicum hemitomon* Schult.) intermingled with bladderworts (*Utricularia gibba* Linn. and *U. inflata* Walt.). False maidencane (*Sacciolepis striata* (Linn.) Nash), arrow-arum (*Peltandra virginica* (Linn.) Kunth) and sedges (*Carex* spp.) were also found in large stands. The onshore vegetation included swamp-bay (*Persea palustris* (Raf.) Sarg.), Florida maple (*Acer barbatum* Michx.), pine (*Pinus* spp.), saw palmetto (*Serenoa repens* (Bartr.) Small), water-primrose (*Ludwigia* spp.) and dog fennel (*Eupatorium capillifolium* (Lam.) Small). Plant terminology follows that of Dressler et al. (1987) and Tarver et al. (1979).

The study site was divided into 70, 25 × 25 m sections along north to south and east to west transects. Each section was subdivided into 4, 12.5 × 12.5 m quadrats (NE, NW, SE and SW). This resulted in 280 quadrats. Samples were collected every 3–5 weeks from approximately 50% of the water-covered quadrats during each sampling period from February 21 to December 17, 1984. On January 15, 1985, samples were

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collected from all water-covered quadrats. Collections were made from the NE and SW quadrats of each section during the first (February 21) and subsequent odd-numbered sampling periods, and from the NW and SE quadrats during the second (April 2) and subsequent even-numbered sampling periods. If one of the preselected quadrats had insufficient water to sample, the nearest available quadrat within the same section was selected.

Water/detritus samples potentially containing larvae were collected from an airboat using the probe and pump system of Morris et al. (1985). The probe was inserted through the vegetation so that the tip was resting on the bottom of the marsh; the pump was turned on for 90 sec. Preliminary work with the system showed that approximately 25 liters of material were pumped in 90 sec (Morris et al. 1985). The water line was cleared before the pump was shut off to prevent carryover of larvae from one sample to another.

Samples were hand sorted for larvae in the laboratory on the day of collection. Larvae were identified to genus and *Cq. perturbans* larvae were tabulated by instar. Water depth and dominant aquatic plant species in the vicinity of the probe were recorded. Water depth and water temperature were also recorded at a baseline station.

Water samples were collected for chemical analysis from 12 randomly selected sites at monthly intervals from June 4 through November 5, 1984. The pH was recorded in the field with a Fisher Accumet model 320 expanded scale research pH meter. Dissolved oxygen was also recorded in the field with a Yellow Springs Instrument model 57 oxygen meter. The remaining water quality parameters were measured in the laboratory. Total alkalinity was determined using the American Public Health Association (APHA) (1981) procedures. Orthophosphate was determined on a Bausch and Lomb Spectronic 20 using methods described by APHA (1981) that were modified for use with prepackaged chemical reagents (Hach Company, P.O. Box 389, Loveland, CO 80539).

The procedures of Norusis (1986, 1988a, 1988b), Taylor (1984) and Elliott (1971) were used to analyze the data on an IBM PC XT computer.

RESULTS

Water chemistry and temperature: Between June 4 and November 5, 1984, the average pH in the study area ranged from 3.6 to 4.9; orthophosphate ranged from <0.001 to 0.14 ppm; total alkalinity from 0 to 5.96 ppm; and dissolved oxygen from 0.6 to 1.4 ppm. The marsh bottom temperature ranged from 10 to 25.5°C.

Seasonal abundance and voltinism: On the 13 sampling dates 15,458 larvae of 5 genera were collected (n = 1,179). Of these, 15,093 (97.6%) were *Cq. perturbans* (Table 1), with an average of 0.72 first instars, 2.18 second instars, 4.46 third instars and 3.42 fourth instars per sample. We also collected 11 pupae, 185 *Uranotaenia* spp., 79 *Anopheles* spp., 66 *Mansonia* spp. and 35 *Culex* spp. These were excluded from further analysis.

The number of winter diapausing fourth instar larvae remaining after early April was low, as indicated by the drop in average instar number on April 24 (Lounibos and Escher 1983) (Table 1). Apparently there was no significant oviposition by adults until after early April. At this time, the number of larvae increased with a concurrent drop in the average instar number. From late April until the end of the study, the average instar number gradually and steadily increased while the number of larvae decreased.

Statistical distributions: For each of the 13 sampling dates the variance exceeded the mean by a factor ranging from 5.6 to 74.7, indicating nonrandomness. The maximum likelihood method (Elliott 1971) of estimating the aggregation coefficient, k, produced values ranging from 0.073 to 0.323. This range of k indicates stability and a potential for using the common k (0.205) to develop sequential sampling methods for population density determination. This study did not address this potential. The chi-square test for "goodness-of-fit" of k to the negative binomial (or log-normal) distribution was accepted for all 13 sampling dates.

Table 1. Summary of *Coquillettidia perturbans* larvae found in 1779 samples collected from a marsh near Lake Alfred, Florida during 1984–85.

Date	No. of samples	*Cq. perturbans* Total	*Cq. perturbans* Mean	Average instar number
Feb. 21	144	119	0.8	3.50
Apr. 2	147	38	0.3	3.92
Apr. 24	145	444	3.1	2.13
May 14	136	2,133	15.7	2.21
June 11	129	1,899	14.7	2.57
July 9	141	1,805	12.8	2.57
Aug. 6	143	1,597	11.2	2.74
Sep. 4	137	1,782	13.0	2.92
Oct. 2	126	708	5.6	2.94
Oct. 29	116	1,294	11.2	3.06
Nov. 26	114	844	7.4	3.59
Dec. 17	107	916	8.6	3.67
Jan. 15	194	1,514	7.8	3.60
Total	1,779	15,093	8.5	2.86

Data were then examined using Taylor's power law (Taylor 1961, Elliott 1971). The value of b in power transformation analysis (slope of the regression of log-variance on log-mean for each sampling date) is an index of aggregation similar to k. For total *Cq. perturbans* larvae, Taylor's b was 2.19 (Table 2, all instars), slightly above the b = 2 expected for a log-normal distribution. The linear regression between log-variance and log-mean of total *Cq. perturbans* was highly significant ($r = 0.97$, $P < 0.001$, $n = 13$).

However, when the appropriate power transformations were calculated for individual instars rather than the total, there was a downward trend in b as larvae aged. This indicates less aggregation in later instars (Table 2). Again, the regressions between variance and mean were highly significant for all instars (Table 2).

Distance from marsh edge: Of the 280 quadrats sampled, 100 (35.7%) were within 25 m of the marsh edge, 115 (41.1%) were in the 26 to 75 m range, and 65 (23.2%) were 76 to 125 m from the edge. Twice as many larvae were found in quadrats located beyond 25 m, but this difference was not significant (Table 3). The frequency distribution of samples containing larvae, based on distance from the edge, was significantly different from all samples (chi-square = 14.7, df = 2, $P = 0.001$). There were more than expected in the 26 to 75 m range (243 vs. 231) and 76 to 125 m range (456 vs. 415) but fewer than expected in the 0 to 25 m range (224 vs. 277).

Relationship to tall vegetation: Early observations suggested that larvae may be more abundant in areas adjacent to trees and shrubs which protrude above the marsh. To test this hypothesis, mosquito production in quadrats within 12.5 m of vegetation taller than 2 m ($n = 162$) was compared to production elsewhere ($n = 118$). Larval density near tall vegetation ($\bar{x} = 9.5$, $n = 953$) was not significantly higher than near shorter vegetation ($\bar{x} = 7.5$, $n = 826$) in ANOVA ($P = 0.41$, $n = 1179$). The frequency distribution of samples that contained larvae, based on proximity to tall vegetation, was not significantly different from the distribution for all samples (chi-square = 0.298, df = 1, $P = 0.585$).

Vegetation: The distribution of samples which contained *Cq. perturbans*, by vegetation, was significantly different from the distribution for all samples (chi-square = 89.5, df = 15, $P < 0.001$). There were more samples with larvae than expected in maidencane (506 vs. 446), arrow-arum (106 vs. 79) and sedge (87 vs. 71). There were fewer positive samples than expected for all but one of the remaining plants (water-primrose, 10 vs. 9). The greatest discrepancies occurred with fragrant water-lily (6 vs. 37), water fern (4 vs. 23), pickerelweed (53 vs. 69) and duckweed (3 vs. 16) quadrats.

There were also significant differences in larval density by vegetation ($P < 0.0001$, ANOVA) (Table 4). The number of differences increased as the larvae aged; 4 differences for first instars, 13 for seconds, 19 for thirds, and 22 for fourths.

Table 2. Power transformation statistics.

	Index of aggregation (Taylor's b)	Power transformation[a]	Mean-variance regression (r)[b]
Cq. perturbans			
1st instars	3.39	$x^{-0.7}$	0.97
2nd instars	2.75	$x^{-0.4}$	0.99
3rd instars	2.63	$x^{-0.3}$	0.98
4th instars	2.44	$x^{-0.2}$	0.92
All instars	2.19	$x^{-0.1}$	0.97
Vegetation			
Miscellaneous	1.36	$x^{0.3}$	0.67
Maidencane	2.14	$x^{-0.1}$	0.98
Sedges	2.16	$x^{-0.1}$	0.97
Arrow-arum	2.22	$x^{-0.1}$	0.96
Pickerelweed	2.99	$x^{-0.5}$	0.97
Water depth (cm)			
<35	2.48	$x^{-0.2}$	0.92
35–55	2.23	$x^{-0.1}$	0.96
60–70	2.09	x^{0}	0.98
>70	2.51	$x^{-0.3}$	0.97

[a] An exponent of: 0.5 is a square-root transformation (Poisson); 0.0 is a logarithm transformation (negative binomial); −0.5 is a reciprocal-of-the-square-root transformation; −1.0 is a reciprocal transformation.

[b] $P < 0.02$ in all cases.

For all instars, arrow-arum, maidencane, false maidencane and sedges had significantly more larvae than 10, 9, 4 and 4 other plants, respectively (Table 5). Bladderworts had significantly more larvae than fragrant water-lily.

Smartweed, spikerush, and red-root had larval densities comparable to arrow-arum, maidencane, sedges and false maidencane (Table 4) but no significant differences were found between these 3 species and other plants. This was due to the very small number of samples and high variability of the larval numbers of the former group compared to the latter group.

Table 3. Density of *Coquillettidia perturbans* larvae by distance from the marsh edge.

Distance from marsh edge (m)	No. of samples	*Cq. perturbans*	
		Arithmetic	Log
		Mean (%)	Mean[a] (%)
0–25	534	4.6 (18.6)	1.2 A (20.3)
26–75	444	10.1 (40.7)	2.2 A (37.3)
76–125	801	10.1 (40.7)	2.5 A (42.4)
Total	1,779	25.6	1.43

[a] Values followed by the same letter are not significantly different, $P = 0.19$ in ANOVA, Student-Newman-Keuls.

For further analysis, each quadrat was assigned to 1 of 5 vegetation communities based on the dominant plant species in that quadrat. The 5 communities were maidencane, arrow-arum, pickerelweed, sedges and heterogeneous areas consisting of various other plant species. Maidencane dominated 120 (42.9%) of the quadrats, arrow-arum dominated 40 (14.3%), pickerelweed 35 (12.5%), sedges 28 (10%) and miscellaneous vegetation 57 (20.4%).

Based on the average number per collection, approximately two-thirds of the larvae were found in the arrow-arum and maidencane areas and one-third in the miscellaneous and *Carex* zones (Table 6). There were no significant differences among the densities in these 4 communities. Less than 3% of the larvae were found in pickerelweed and the density was significantly lower than in the other four groups (Table 6).

Grouping quadrats by vegetation community permitted us to also quantify the spatial distributions by vegetation. Larval distributions were log-normal in arrow-arum, maidencane and

Table 4. Density of *Coquillettidia perturbans* larvae in 1,779 samples dominated by 22 vegetation types and in open water.

Vegetation	No. of samples	Mean number of *Cq. perturbans*	
		Arithmetic	Log
Best			
Arrow-arum (*Peltandra virginica*)	151	13.8	5.5
Maidencane (*Panicum hemitomon*)	856	12.2	3.8
Smartweed (*Polygonum hydropiperoides*)	11	14.5	3.7
Good			
Red-root (*Lachnanthes caroliniana*)	9	5.6	2.8
False maidencane (*Sacciolepis striata*)	164	5.5	2.7
Sedges (*Carex* spp.)	137	3.6	2.7
Spikerush (*Eleocharis baldwinii*)	19	6.1	2.7
Fair			
Bladderworts (*Utricularia* spp.)	66	6.5	2.3
Water-primroses (*Ludwigia* spp.)	18	3.3	2.3
Spatterdock (*Nuphar luteum*)	10	2.7	1.9
Pickerelweed (*Pontederia cordata*)	132	2.0	1.7
Water pennywort (*Hydrocotyle umbellata*)	25	2.3	1.3
Poor			
Swamp-bay (*Persea palustris*)	2	0.5	1.4
Saw grass (*Cladium jamaicense*)	2	0.5	1.4
Buttonbush (*Cephalanthus occidentalis*)	17	0.5	1.3
Water fern (*Salvinia rotundifolia*)	45	0.7	1.2
Common duckweed (*Lemna minor*)	30	0.2	1.1
Fragrant water-lily (*Nymphaea odorata*)	72	0.2	1.1
Mosquito fern (*Azolla caroliniana*)	2	0.0	0.0
Bog-mats (*Wolffiella* spp.)	2	0.0	0.0
Brazilian-pepper (*Schinus terebinthifolius*)	2	0.0	0.0
Ferns (*Pteridophytes* other than *Salviniacea*)	5	0.0	0.0
Open water	1	0.0	0.0
Unknown	1	0.0	0.0

Table 5. Pairs of plant species with significantly ($P < 0.05$, ANOVA-SNK) different populations of larval *Coquillettidia perturbans*.

	Arrow-arum	Maidencane	False maidencane	Sedges	Bladderworts
Fragrant water lily	2,3,4,A[a]	1,2,3,4,A	3,4,A	3,4,A	A
Duckweed	2,3,4,A	2,3,4,A	A	A	
Water fern	2,3,4,A	2,3,4,A	4,A	4,A	
Pickerelweed	3,4,A	1,2,3,4,A	4,A	4,A	
Sedges	2,3,4,A	1,2,3,4,A			
False maidencane	2,3,4,A	1,2,3,4,A			
Bladderworts	2,3,4,A	3,4,A			
Water pennyworts	2,3,4,A	3 A			
Buttonbush	3,4,A	A			
Maidencane	A				

[a] 1, 2, 3 and 4 = larval instar number, A = all instars.

Table 6. Density of *Coquillettidia perturbans* larvae, by major vegetation categories.

		Cq. perturbans			
		Arithmetic		Log	
Vegetation	No. of samples	Mean	(%)	Mean[a]	(%)
Arrow-arum	256	11.8	(33)	3.3 A	(34)
Maidencane	816	11.9	(34)	2.9 A	(30)
Miscellaneous	283	7.8	(22)	1.8 A	(19)
Sedges	183	3.0	(8)	1.3 A	(14)
Pickerelweed	241	1.0	(3)	0.3 B	(3)
Total	1,779	35.5		2.2	

[a] Values followed by the same letter are not significantly different, $P = 0.0001$, Oneway ANOVA, Student-Newman-Keuls.

sedge dominated communities, even more aggragated in pickerelweed, but were nearly random in miscellaneous vegetation areas (Table 2).

Water depth: Mean water depth ranged from 31 to 52 cm and changed primarily in response to rainfall patterns. The preadult stages of *Cq. perturbans* develop slowly and, apparently, based on this study, can take up to a year to develop. Assuming larvae migrated little from sites of oviposition, the immatures experienced widely fluctuating water levels during their development. Therefore, it was necessary to analyze the relationship between *Cq. perturbans* abundance and water depth in 2 ways. The first was to examine abundance based on the water depth when the sample was taken. In the second analysis each quadrat was assigned to one of 4 water depth groups based on its seasonal average depth. This permitted the comparison across these groups and the quantification of spatial distributions in waters of various depths.

Larvae were abundant in 35 to 70 cm deep water, with 60–70 cm being optimum (Table 7). There were significantly fewer larvae in areas with less than 35 cm of water (Table 8). Based

Table 7. Density of *Coquillettidia perturbans* larvae by water depth when the sample was taken.

Water depth (cm)	No. of samples	Mean number of *Cq. perturbans*	
		Arithmetic	Log
10	8	0.0	0.0
15	12	0.0	0.0
20	29	0.3	0.2
25	55	0.2	0.1
30	24	0.9	0.2
35	33	7.0	1.4
40	45	6.2	1.4
45	53	4.7	1.3
50	61	6.4	2.0
55	90	8.9	1.6
60	121	10.9	3.1
65	273	14.7	3.8
70	363	13.0	3.2
75	360	5.6	1.5
80	180	4.0	1.3
85	50	1.9	1.1
90	0	—	—
95	17	1.5	1.0
100	0	—	—
105	5	0.0	0.0
Total	1,779		

Table 8. Density of *Coquillettidia perturbans* larvae by water depth categories.

		Density of *Cq. perturbans*			
		Arithmetic		Log	
Water depth (cm)	No. of samples	Mean	(%)	Mean[a]	(%)
<35	156	0.6	(2)	1.2 A	(11)
35–55	217	9.2	(33)	2.9 B	(27)
60–70	744	13.8	(50)	4.5 B	(41)
>70	662	4.1	(15)	2.3 B	(21)
Total	1,779	34.2		3.0	

[a] Values with the same letter are not significantly different, $P = 0.0001$, oneway ANOVA, Student-Newman-Keuls.

on the frequencies of all samples collected, there were also substantially fewer samples with larvae than expected in the 0 to 35 cm range (19 vs. 81) but substantially more than expected in the 60 to 70 cm range (477 vs. 386) (chi-square = 71.1, df = 3, $P < 0.0001$).

DISCUSSION

Water chemistry: The low pH and dissolved oxygen conditions observed in this study are typical for *Coquillettidia* breeding sites in central Florida (Lounibos and Escher 1983, Callahan and Morris 1987a), elsewhere in North America (Allan et al. 1981, Batzer and Sjogren 1986, and others) and in southern France (Guille 1976).

Seasonal abundance and voltinism: In Florida, the annual number of generations of *Cq. perturbans* typically ranges from one in the temperate north to 3 in the tropical south (Provost 1976). While there can be both spring and fall emergences in central Florida (Provost 1976, Lounibos and Escher 1983), in 1984 only the spring brood was apparent at the Lake Alfred site.

Theoretically, assuming equal sampling efficiency for all instars, the lowest average instar number should occur simultaneously with the highest number of larvae. This was not the case in our study. The lowest average instar number (2.13) occurred in late April, one sampling period before peak numbers of larvae in May. This seems higher than expected considering that 3 weeks before (April 2), 92% of the larvae collected were fourth instars and yet, on May 14, 3 weeks after the average instar number low point, the average instar number had risen only 3.8% (Table 1). These observations suggest that either larvae remained first instars for only a short period of time or first instar larvae were underrepresented in the collections.

The 350 μm mesh screen used in the probe and pump system was larger than the 260 to 300 μm head capsule widths of first instar *Cq. perturbans* (Nemjo and Slaff 1984). This would allow some first instars to pass through the sieve. Head capsule widths of second to fourth instar larvae in the Nemjo and Slaff study all exceeded 410 μm; theoretically, all of these larvae would have been collected. Field trials of screens with mesh size less than 350 μm resulted in unacceptable plugging problems. The lower than expected numbers of first instar larvae could also be attributed to the greater difficulty in finding these small, transparent insects in the often murky collection water. This collection bias means the average instar number in Table 1 is an overestimate of age, at least through mid-July while first instars were still abundant.

The depression in both larval numbers and the rate of increase of the average instar number in early October may indicate a minor fall emergence that occasionally occurs in central Florida (Lounibos and Escher 1983). This seems unlikely since there was a decrease in numbers of all instars.

Statistical distributions: The most appropriate transformation method for *Cq. perturbans* larval data would be the power law on an instar by instar basis. That is, replace each instar count by X^p, where $p = 1 - (b/2)$ and b is Taylor's b for that instar. The b values observed, however, were only slightly greater than the b value of a negative binomial (i.e., 2). For practical purposes, *Cq. perturbans* larvae are distributed approximately as a negative binomial; therefore, sample numbers can be normalized using log transformations. This practice should already be routine when analyzing insect trapping data (Wadley 1967, Steel and Torrie 1960).

from the egg raft from which they hatched. The relative magnitude of the changes in b between instars suggests that essentially all of the dispersion occurred in the second instar. One could argue that, since the index never approached that of a random distribution (i.e., 1), *Cq. perturbans* larvae must have dispersed only short distances.

Distance from marsh edge: There were more larvae, and more samples with larvae than expected, in the 76 to 125 m zone, but fewer than expected in the 0 to 25 m zone. Using the rationale of the edge effect theory, one would expect greater numbers of larvae in the 0 to 25 m zone. Guille (1976) explained his lower densities at the edge of his marsh as the result of unsuitably high soil temperatures produced by evaporation and drought drying out the edge. Lounibos and Escher (1983) found equal numbers of *Cq. perturbans* near and away from the edge of a vegetation covered phosphate pit which had steep sloped edges not susceptible to drying out.

Vegetation: There was no evidence in this study that tall vegetation either in the center or at the edge of the marsh influenced oviposition site selection. In a later study Callahan and Morris (1987a) found that mosquito larvae of 5 genera, including *Coquillettidia*, were more abundant in lake waters adjacent to tall shoreline vegetation that apparently served as diurnal resting refuges for gravid adults. In an earlier study on the distribution of *Mansonia dyari* (Belkin, Heinemann and Page) and *Ma. titillans*

(Walker) larvae on water lettuce (*Pistia stratiotes* Linn.), we also observed higher larval densities in waters adjacent to tall shoreline vegetation (Morris et al. 1986).

Arrow-arum, maidencane and smartweed make up a group of plant species that were indicators for the "best" habitats for *Cq. perturbans* larval development and survival (Table 4). Redroot, false maidencane, sedges and spikerush were indicators of "good" habitats. Communities dominated by these 7 species typically had very dense vegetation, often with a floating mat component. The subsurface structure also often included submergent plant species.

Bladderworts, water-primroses, spatterdock, pickerelweed and water pennywort supported only "fair" larval populations. Mosquitoes were seldom found in monotypic stands of bladderworts, a submerged plant, but were often abundant where bladderworts were mixed with one or more emergent plants. In these cases bladderworts may have provided larvae additional protection from predation, even though bladderworts can themselves be larvivorous (Baumgartner 1987). Dense mats of bladderwort reduce not only predation, but also gravid females' access to the water surface for oviposition, and fewer larval attachment sites.

In contrast, spatterdock and pickerelweed occurred in more sparsely populated monotypic stands that did not exclude predators. Larvae found associated with water-primroses were probably from eggs laid near other plants since water-primroses do not possess appropriate roots for larval attachment.

The 10 plant species in the last group indicated "poor" larval survival areas and were either 1) woody plants without suitable larval attachment sites (swampbay, buttonbush and Brazilian-pepper); 2) small floating plants which tend to exclude oviposition (water fern, duckweed, mosquito fern and bog-mat) (Hobbs and Molina 1983); 3) monocultures in deep waters subject to wave action and/or which allow easy access by predators (saw grass, fragrant waterlily, pickerelweed and spatterdock); or 4) were temporarily inundated terrestrial non-Salviniaceae ferns located at the edge of the marsh which tended to dry up.

In general, *Cq. perturbans* larvae were most aggregated in "fair" plant communities, and log-normally distributed in "best" and "good" communities. The near randomness observed in the miscellaneous plant zones probably resulted from the mosaic of plant types which were themselves more or less randomly distributed.

We concur with Guille's (1976) observations on *Cq. richiardii* (Ficalbi) that higher density and greater diversity of vegetation provide the best larval habitats. These habitats provide obvious barriers to predators, especially fish.

While plant species can be a major parameter to use in characterizing the breeding potential of a marsh or other vegetated wetland, caution is advised. For example, in this study, smartweed was classified in the "best" category. Yet, in a preliminary study of several lake littoral zones, and in a subsequent study of 2 marshes and 2 additional lake littoral zones, we never found larvae associated with this plant (Callahan and Morris 1987b). The absence of a detrital layer in the latter cases precluded larval survival.

Pickerelweed was classified as only a "fair" indicator of abundance in the current study. However, Bidlingmayer (1950) found pickerelweed to be one of the "best" hosts in a marsh in Leesburgh, FL, just 50 miles north of our Lake Alfred site.

A third case in point pertains to *Cq. perturbans* larvae which are highly associated with cattails (*Typha* spp.), particularly in temperate climates. Throughout its range, *Cq. perturbans* larval densities are higher on floating mats of cattails than on rooted plants (Batzer and Sjogren 1986, Slaff and Haefner 1985).

One is well advised to heed the words of Robert Armstrong (1941) who wrote: "In searching for breeding places [of *Mansonia perturbans*], it is better to be guided by the appearance and texture of the marsh than by a list of host plants."

Water depth: Our findings of larval density in relation to water depth agrees precisely with Guille (1976), who also found the greatest densities of *Cq. richiardii* in the 60 to 70 cm range. He also found less than half as many at depths below 30 cm and above 70 cm, although he explained the lower densities at the greater depths as an artifact of his collection method. Since the collection efficiency of our sampling method was independent of water depth and we also found progressively fewer larvae at greater depths, we do not believe Guille's results are erroneous.

Based on Taylor's index of aggregation, larvae were log-normally distributed in the optimum water depth zones (35 to 70 cm) and slightly more aggregated in less favorable depths (Table 2); a trend similar to that seen earlier with vegetation.

CONCLUSION

While larval aggregation was related to vegetation, water depth and proximity to the marsh edge, but not proximity to tall vegetation, the aggregation was insufficient to pinpoint control efforts such as is done in Minnesota. However,

some control could be achieved by managing shore and aquatic vegetation, and water depth.

While this approach may not be possible in natural marshes, it is possible in manmade, restored and enhanced wetlands projects, including lake shores. Such activities are on the increase in Florida and elsewhere, and environmental engineers are quickly learning how to design and build productive marshes. Unfortunately, designs are often unintentionally ideal for the mass production of mosquitoes.

Vegetation and water depth can be easily manipulated in storm and waste water retention and detention ponds. These facilities are often designed and constructed in urban areas without considering the mosquito producing potential.

Wildlife management areas often include wetlands ideal for *Cq. perturbans* production. Management plans for these wetlands, when managed for waterfowl, usually include lists of desirable vegetation species, slope ratios and water depths. Making up "desirable plant" lists should also take mosquito breeding potential into consideration. The incidence of mosquito transmitted wildlife diseases such as avian malaria and EEE may also be reduced if mosquitoes in these areas are reduced.

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