Host Age Effects on Ovipositional and Developmental Biology of *Baryscapus chrysopae* (Hymenoptera: Eulophidae), a Parasitoid of Chrysopid Larvae[1]

John R. Ruberson[2] and Timothy J. Kring

Department of Entomology, University of Arkansas,
Fayetteville, Arkansas 72701 USA

J. Entomol. Sci. 30(2): 287-293 (April 1995)

ABSTRACT The parasitoid, *Baryscapus* (=*Tetrastichus*) *chrysopae* (Crawford), is a widely-distributed gregarious parasitoid of chrysopid larvae. The ovipositional and developmental biology of this parasitoid in relation to the stage of its host, *Chrysoperla rufilabris* (Burmeister), was examined. Female *B. chrysopae* attacked all larval stages of the host tested (instars 1 to 3), and paralyzed all hosts soon after mounting and stinging. During the host's paralysis, females oviposited in and fed on hosts. The time females spent on hosts was directly related to host stage. All hosts recovered from paralysis. Parasitoid developmental time was inversely related to host stage and ranged from 27.5 d in 1-d-old hosts to 20.5 d in 10-d-old hosts. Most development of parasitoid larvae appears to occur after the host has spun its pupal cocoon. The number of parasitoids produced per host was unrelated to host stage, ranging from 10.5 (in 1-d-old hosts) to 14.2 (in 7-d-old hosts) parasitoids per host. The sex ratio was skewed toward females (81.6% pooled across host stages) and was unrelated to host stage. The developmental biology of *B. chrysopae* appears to be well synchronized with that of its host.

KEY WORDS Insecta, *Baryscapus chrysopae, Chrysoperla rufilabris,* parasitoid, predator, Neuroptera, lacewing, parasitoid behavior, host age.

Larvae of chrysopids are attacked by a number of hymenopteran parasitoids (Clancy 1946, Principi 1948, Neumark 1952, Muma 1959, Alrouechdi et al. 1984). This complex of parasitoids can reduce populations of lacewings, thus interfering with their efficacy as biological control agents (McGregor 1914, Smith 1922, Putman 1937, Clancy 1946, Alrouechdi et al. 1981, Gerling and Bar 1985). The gregarious larval parasitoid *Baryscapus* (formerly *Tetrastichus*) *chrysopae* (Crawford) is a commonly-occurring natural enemy of chrysopids in the Holarctic region. Clancy (1946) and Neumark (1952) reviewed the general biology of this parasitoid in California and Israel, respectively.

[1] Accepted for publication 04 January 1995.

[2] Current address: Dept. of Entomology, Univ. of Georgia, Coastal Plain Exper. Sta., P. O. Box 748, Tifton, GA 31793.

Because of the widespread distribution of this parasitoid and the importance of chrysopids in biological control, studies addressing the biology of *B. chrysopae* may provide valuable insights into its full impact on chrysopid populations. In the present study we examined the ability of *B. chrysopae* to parasitize and develop in various larval stages of the economically-important common green lacewing, *Chrysoperla rufilabris* (Burmeister).

Materials and Methods

Acquisition and Rearing of Parasitoids. In August 1991, parasitoids were reared from 12 chrysopid cocoons collected from a cotton field in Little River Co., AR. All of these parasitoids were determined to be *B. chrysopae* by J. LaSalle (British Museum of Natural History). They differ, however, from the parasitoids studied by Clancy (1946) and Neumark (1952): the parasitoids we studied are entirely black, whereas the populations studied by Clancy (1946) and Neumark (1952) were reported to have testaceous legs and metallic-green gasters.

After parasitoid adults emerged from the chrysopid cocoons, they were paired, and individual pairs (siblings were paired; we assumed sib-mating on the basis of the gregarious habit of this parasitoid) were held in vials (provisioned with honey) at 25 ± 1°C, L:D = 14:10. Host larvae for the experiments were reared from eggs collected in cotton maintained on the Arkansas Agricultural Experiment Station Research Farm (Fayetteville, AR). Chrysopid larvae were identified as *C. rufilabris* using the larval keys of Tauber (1974) and Agnew et al. (1981). Voucher specimens of the parasitoids have been placed in the University of Arkansas Arthropod Museum.

Experimental Procedure. To evaluate acceptance of various larval host stages of their hosts and to observe ovipositional behavior of the parasitoid, individual mated female *B. chrysopae* were placed in a Petri dish (15 × 60 mm) with a single host larva of the selected stage (see stages listed in Table 1). Each female parasitoid was used once; 20 to 36 *C. rufilabris* larvae were stung for each larval stage tested. Behaviors of the female parasitoid and the host were noted and timed. A host larva was considered paralyzed when it no longer responded to gentle prodding with a probe. Parasitoids were removed from the dish after leaving the host. Stung host larvae were held in 30-ml diet cups at 25 ± 1°C, L:D = 14:10, and provided with field-collected corn leaf aphids (*Rhopalosiphum maidis* L.) as prey until the larva spun its pupal cocoon or died. All host cocoons and larval remains were dissected ≥ 35 d after stinging (after expected parasitoid emergence from the host) to determine the number of parasitoids that had failed to emerge. Unemerged parasitoids were sexed and added to the number of parasitoids emerged to obtain a corrected sex ratio and number of parasitoids developing per host.

Statistical Analyses. Effect of host stage on the time to, and duration of, host paralysis, as well as the amount of time parasitoids spent on hosts, were examined using one-way analysis of variance (ANOVA) (SAS Institute 1985). When differences were significant, means were separated using the Waller-Duncan Bayesian k ratio (Waller and Duncan 1969). Parasitoid developmental times and number of parasitoids developing per host in relation to host stage also were analyzed with

ANOVA, followed by means separation with the Waller-Duncan Bayesian k ratio when significance was demonstrated. Sex ratio of parasitoid offispring in relation to host stage were evaluated with a G test (Sokal and Rohlf 1981).

Results and Discussion

Female *B. chrysopae* readily attacked host larvae of all stages tested. Parasitoids usually mounted hosts on the dorsum (Fig. 1), but they occasionally attacked on the venter in the host's thoracic region. First instar hosts were mounted anywhere posteriad of the head. When hosts were second or third instars, however, parasitoids concentrated their attacks on the cervical area. Clancy (1946) and Neumark (1952) observed a similar preference for the dorsum in their studies of *Tetrastichus* spp. with the chrysopid *C. carnea* (Stephens).

After mounting the host, the parasitoid quickly inserted its ovipositor, after which the host larva ran about in the dish and attempted to dislodge the parasitoid by arching its head back toward the parasitoid and attempting to grasp the stinging female with its long mandibles. In one case, a female parasitoid mounted a third-instar host on the larva's metathoracic dorsum and was seized and killed by the larva. All other females restricted their attacks to the cervical area.

After being mounted and stung, *C. rufilabris* larvae succumbed rapidly to paralysis (Table 1). The time from mounting to paralysis was consistent (range 1-4 min) and unrelated to host instar. Clancy (1946) found that slowing of the host in his studies did not occur until 5-10 min after parasitoid stinging, and paralysis followed several minutes later. The parasitoids in the present study were thus able to subdue hosts much more rapidly than those studied by Clancy (1946).

Duration of the paralysis was related to host instar; third instars were paralyzed for significantly longer periods than were first and second instars (Table 1). In all cases, paralysis of larvae in our study was considerably longer than the 12 to 21 min durations reported by Clancy (1946). All paralyzed larvae appeared to recover completely. Clancy (1946) found that hosts resumed activity approximately 5 min after departure of the parasitoids, whereas in our study, time to activity resumption varied with host stage (Table 1), but was always more than 15 min after parasitoid departure.

The amount of time parasitoids spent on hosts was directly related to host stage (Table 1). Only two activities were observed while the parasitoids were on the host: stinging and host feeding. Host feeding at the stinging site followed stinging in all cases (Fig. 1). Duration of host feeding was not recorded in these studies. Likewise, Clancy (1946) observed feeding by all ovipositing females. After feeding was completed, female parasitoids left the host larvae.

Developmental times of parasitoid offspring were inversely related to host stage (Table 2). However, the developmental time required for parasitoids after the host spun its cocoon was comparable for all host stages except 10-d-old larvae (third instars; Table 2), which was significantly longer than the other stages. These data support Clancy's (1946) assertion that larvae of *B. chrysopae* in younger host larvae delay development until the host reaches a suitable stage for continued parasitoid development. He observed that development of *B. chrysopae* beyond the second instar was suppressed until the host had spun its cocoon. This suppression of development may be important for sychronizing the parasitoid with the phenology of its host.

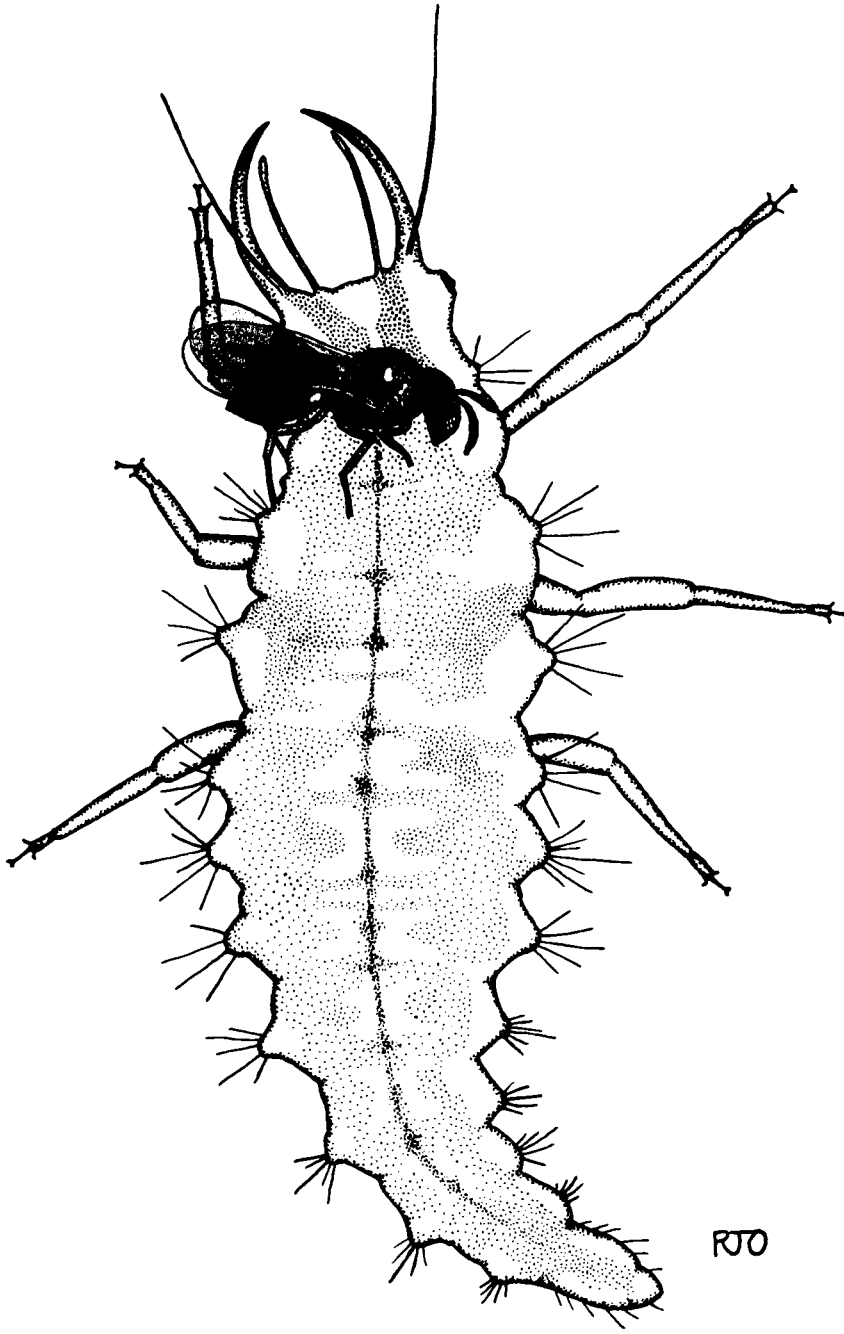

Fig. 1. *Baryscapus chrysopae* female host feeding on 3rd instar larva of the chrysopid *Chrysoperla rufilabris.* Drawing (from photograph) by Russell J. Ottens, Univ. of Georgia.

Table 1. Relationship between age of the host (*Chrysoperla rufilabris*), host paralysis and time spent on host by the parasitoid *Baryscapus chrysopae* (mean ± SD)*

Host instar (age, d)	n	Min to host paralysis	Duration of paralysis	Min spent on host
1 (1 d)	26	1.8 ± 0.37 a	52.3 ± 3.61 a	14.4 ± 0.72 a
2 (3 d)	20	2.4 ± 0.79 a	51.7 ± 4.61 a	25.3 ± 4.23 b
3 (7 d)	36	1.9 ± 0.65 a	68.6 ± 11.99 c	28.7 ± 1.61 b
3 (10 d)	30	2.3 ± 0.54 a	58.1 ± 10.61 b	43.3 ± 6.60 c

* Values in columns followed by the same letter are not significantly different (Waller-Duncan Bayesian K ratio, $K = 100$).

The number of parasitoids developing per host was not significantly affected by the host stage (Table 2). Nor did the sex ratio of the offspring differ significantly in response to host stage (Table 2). Sex ratio was strongly female-biased with a pooled average of 81.6 ± 12.23% females per brood. These results are similar to those of other tetrastichine parasitoids of chrysopid larvae (Alrouechdi et al. 1984). Clancy (1946) noted an average of 12.3 *B. chrysopae* emerging per host with 75.6% of them female. Neumark (1952) indicated that 10-26 parasitoids emerged from each host ". . . with a slight preponderance of females."

Nearly all parasitized host larvae were killed by emerging parasitoids while they were prepupae in the cocoon as Clancy (1946) observed. Six parasitized host larvae, however, died as third instars, before spinning their cocoons, with numerous parasitoid larvae in them. Parasitoid larvae emerged from two of these cadavers and pupated on the floor of the rearing cup two days after emerging. All of these parasitoid pupae subsequently produced adult parasitoids. In addition, three chrysopid larvae that had been stung produced apparently healthy adult lacewings. Whether parasitoid eggs had been encapsulated or the parasitoids had simply failed to oviposit in these hosts is unknown.

The developmental biology of *B. chrysopae* permits close synchrony with host development. *C. rufilabris* develops through the pupal stage in about 10 d at 25°C, then requires an approximately 4 d preoviposition period (Putman 1937). Thus, adult parasitoids emerge from hosts during a period when adult lacewings of the same larval generation as that which had been parasitized are ovipositing; therefore, hosts will be available for parasitism when adult parasitoids emerge. In addition, adult *B. chrysopae* are also long-lived; females in the laboratory lived 1-2 months (J.R.R., pers. obs.). Clancy (1946) likewise observed adult longevity of 30-70 days in the laboratory. Therefore, these parasitoids are well synchronized with their hosts in both their development and longevity.

B. chrysopae possesses several attributes that would allow it to reduce chrysopid populations: the capacity to attack and develop in a range of host stages and developmental synchrony with hosts. However, the parasitoid's seasonal synchrony with its

Table 2. Effects of host (*Chrysoperla rufilabris*) age on development (24 ± 1 C, L:D = 14:10) and sex ratio of the parasitoid *Baryscapus chrysopae* (mean ± SD)*

Host instar (age, d)	n	Developmental time (d)**	Time after host spinning (d)	No. parasitoids per host	Parasitoid sex ratio (%)
1 (1 d)	118	27.8 ± 2.66 a	13.8 ± 1.13 a	10.5 ± 2.07 a	74.9 ± 10.75
2 (3 d)	172	25.3 ± 2.17 b	14.5 ± 2.16 a	11.8 ± 3.19 a	65.4 ± 12.32
3 (7 d)	318	21.9 ± 2.01 c	14.8 ± 1.32 a	14.2 ± 5.37 a	85.2 ± 10.69
3 (10 d)	160	20.5 ± 0.64 c	18.0 ± 2.29 b	12.6 ± 3.66 a	87.8 ± 8.43

* Values in columns followed by the same letter are not significantly different (Waller Duncan Bayesian K ratio, $K = 100$). Sex ratios, examined with G test, did not differ significantly with host age.

** Time from parasitization to adult parasitoid emergence.

hosts, its capacity to locate hosts in the field, the range of chrysopid species it is capable of utilizing, and its response to varying densities of chrysopid larvae are unexplored. Studies in these areas would provide valuable insights into the dynamics of chrysopid populations in the field.

Acknowledgment

We appreciate the expeditious determination of the parasitoid by J. LaSalle (British Museum of Natural History, London) and the comments of D. C. Steinkraus and J. B. Whitfield (Univ. of Arkansas), and W. L. Tedders (USDA-ARS) on the manuscript. We also appreciate the assistance of L. Bush with the experiments. This research was supported by USDA-Cooperative States Research Service Grant 89-34195-4378. This article is published with the approval of the director, Arkansas Agricultural Experiment Station, University of Arkansas, Fayetteville.

References Cited

Agnew, C. W., W. L. Sterling and D. A. Dean. 1981. Notes on the Chrysopidae and Hemerobiidae of eastern Texas with keys for their identification. Southwest. Entomol. Suppl. No. 4.

Alrouechdi, K., Y. Semeria and T. R. New. 1984. Natural enemies. *In* M. Canard, Y. Semeria, T. R. New (eds.), Biology of Chrysopidae, pp. 187-193. Junk, The Hague.

Alrouechdi, K., M. Canard, R. Pralavario and Y. Arambourg. 1981. Influence du complexe parasitaire sur les populations de Chrysopides (Neuroptera) dans un verger d'oliviers du Sud-Est de la France. Z. ang. Entomol. 91: 411-417.

Clancy, D. W. 1946. The insect parasites of the Chrysopidae (Neuroptera). Univ. Calif. Publ. Entomol. 7: 403-496.

Gerling, D. and D. Bar. 1985. Parasitization of *Chrysoperla carnea* (Neuroptera, Chrysopidae) in cotton fields of Israel. Entomophaga 30: 409-414.

McGregor, E. A. 1914. Some notes on parasitism of chrysopids in South Carolina. Can. Entomol. 46: 306-308.

Muma, M. H. 1959. Hymenopterous parasites of Chrysopidae on Florida citrus. Flor. Entomol. 42: 149-153.

Neumark, S. 1952. *Chrysopa carnea* St. and its enemies in Israel. Ph.D. dissertation, Hebrew University, Ilanoth. 136 pages.

Principi, M. M. 1948. Contributi allo studio dei Neurotteri italiani. VII. Osservazioni sul alcuni parassiti di Crisopidi. Boll. Ist. Entomol. Univ. Bologna 17: 93-121.

Putman, W. L. 1937. Biological notes on the Chrysopidae. Can. J. Res., Sec. D 15: 29-37.

SAS Institute. 1985. SAS User's Guide: Statistics. SAS Institute, Inc., Cary NC.

Smith, R. C. 1922. The biology of the Chrysopidae. Cornell Univ. Agric. Exp. Sta. Mem. 58: 1287-1372.

Sokal, R. R. and F. J. Rohlf. 1981. Biometry, 2nd Ed. Freeman and Co., NY.

Tauber, C. A. 1974. Systematics of North American chrysopid larvae: *Chrysopa carnea* group (Neuroptera). Can. Entomol. 106: 1133-1153.

Waller, R. A. and D. B. Duncan. 1969. A Bayes rule for the symmetric multiple comparisons problem. J. Am. Stat. Assoc. 64: 1484-1503.