Zelus renardii Roaming in Southern Italy

Nada Lahbib 1,2, Ugo Picciotti 1,4,* Valdete Sefa 1, Sonia Boukhris-Bouhachem 3, Francesco Porcelli 1,5 and Francesca Garganese 1

1 Dipartimento di Scienze del Suolo, della Pianta e degli Alimenti, University of Bari Aldo Moro, 70126 Bari, Italy; nadalahbib48@gmail.com (L.N.); ugo.picciotti@uniba.it (P.U.); valdetesefa@gmail.com (S.V.); francesco.porcelli@uniba.it (P.F.); francesca.garganese@uniba.it (G.F.)
2 Faculty of Sciences of Tunis, University of Tunis El-Manar, Rommana, Tunis 1068, Tunisia
3 Laboratory of Plant Protection, INRAT—National Institute of Agronomic Research of Tunisia, Carthage University, Ariana 2049, Tunisia; bouhachems@gmail.com (B.-B.S.)
4 Laboratory of Phytopathology, Department of Marine Science and Applied Biology, University of Alicante, San Vicente del Raspeig, 03690 Alicante, Spain
5 CIHEAM-Centre International de Hautes Etudes Agronomiques Méditerranéennes, Mediterranean Agronomic Institute of Bari, 70010 Valenzano, Italy
* Correspondence: ugo.picciotti@uniba.it

Simple Summary: The leafhopper assassin bug Zelus renardii (Hemiptera: Reduviidae) first entered Europe in 2012 and has since acclimatised. Z. renardii is now a common insect predator in agroecosystems and urban and peri-urban areas. We performed a metadata analysis of 165 years of Z. renardii literature. Moreover, we provide laboratory tests of Z. renardii predation. The latter indicates Z. renardii interplay with relevant insect targets, such as Philaenus spumarius, Neophilaenus campestris, Bactrocera oleae, Kermes vermilio, Nidularia pulvinata, Harmonia axyridis, Apis mellifera, Aleurocanthus spiniferus, Aleurothrixus floccosus, Macrophomotoma gladiata, Drosophila suzukii, Drosophila melanogaster, Megaselia scalaris, Pseudococcidae, Miridae, and Issidae. Furthermore, predation on Aphrophoridae and other olive pests brands Z. renardii as a good mass-rearing candidate for inundative biocontrol programs of Xylella fastidiosa pauca ST53 infections and could also reduce damage caused by other olive pests. Overall, this reduviid is harmless to humans and beneficial insects.

Abstract: This study collects data from the literature and updates our Zelus renardii Kolenati, 1856 (Leafhopper Assassin Bug, LAB) prey knowledge. The literature consists of ca. 170 entries encompassing the years 1856 to 2021. This reduviid originated in the Nearctic region, but has entered and acclimatised in many Mediterranean countries. Our quantitative predation experiments—in the laboratory on caged plants plus field or environmental observations—confirm that LAB prefers a selected array of prey. Laboratory predation tests on living targets (Hemiptera, Coleoptera, Diptera, and Hymenoptera) agree with the literature. Zelus renardii prefers comparatively large, highly mobile, and readily available prey. LAB preferences on available hemipterans targets suggest that Zelus renardii is a good inundative biocontrol agent for Xylella fastidiosa pauca ST53 infections. LAB also prey on other important olive pests, such as Bactrocera oleae. Therefore, Zelus renardii is a major integrated pest management (IPM) component to limit Xylella fastidiosa pandemics and other pest invasions.

Keywords: alien invasive or quarantine; stenophagous predator; infection and vector biocontrol; Harpactorinae; reference metadata analysis

1. Introduction

Studies on Macrohomotoma gladiata (Kuwayama, 1908) [1] and Aleurocanthus spiniferus (Quaintance, 1903) in Apulia [2] disclosed the acclimation of the Leafhopper Assassin Bug (LAB) or Zelus renardii Kolenati, 1856 (Reduviidae: Harpactorinae—Zr) in southern Italian areas threatened by Xylella fastidiosa (Wells, Raju et al., 1986—Xf) pauca ST53 invasion [3,4].
The first observations of the LAB attacking and killing abilities versus *Philaenus spumarius* (L., 1758—meadow spittlebug) [5,6] and *Neophilaenus campestris* (Fallén, 1805) suggest LAB as a biocontrol agent of Xf vectors. *Philaenus spumarius* is the primary Xf vector in Italy with a 25–71% transmission efficiency [7,8]. *Philaenus spumarius* causes much of the olive (*Olea europaea* L., 1753) infections and the invasive spread of Xf over territories [9].

Expansion of olive orchards under organic practices requires new crop protection strategies to manage the transmission by Xf vectors [10,11]. LAB’s bionomics could fit the needs of organic olive pest control by using the mass-reared reduviid as a biocontrol agent against Xf vectors and other pests. However, this proposal has been met with various opinions and concerns.

LAB is considered a generalist predator [12–17]. However, even generalist predators show prey preferences that satisfy their developmental and reproductive needs [18]. The range of prey depends on physiological, physical, and behavioural limitations [19]. Accordingly, the prey list of LABs in the field usually has fewer species than those reported in the literature, as for other generalist predators [18–21].

This contribution contains two sections: the first deals with available LAB references and their content; the second includes experimental data from laboratory tests, describing LAB’s actions versus relevant prey.

We aim to share the results of the *Z. renardii* tests carried out in Italy. These tests evaluate if LAB uses Italian prey vicariant of the North American ones or beneficial insects as targets such as honeybees. The data will help clarify LAB’s role as a potential biocontrol agent of Xf vectors and infection.

2. Materials and Methods

2.1. LAB References Retrieval

We retrieved LAB references from several repositories, using the systematic review method (Figure 1). The bibliography was consulted in two steps using an exact string, namely “*Zelus renardii*”. The first step is an initial search on databases for the 1856–2020 interval; the second step is to search for bibliographic updates published in 2021, one year later, in the same databases. Databases selected were CAB-Abstract via Ovid, Crop Protection Compendium® (CPC) and Google Scholar. The period from 1856 to 2021 was analysed.
Figure 1. The process adopted to select LAB references analysis, used in this study.

A further nested search into each publication’s references list gathered some additional relevant articles that were included in this study.

2.2. LAB Field Collection

Sweeping nets, beating trays, and direct plant observations served to collect LAB adults, nymphs, and egg batches. Table 1 shows LAB collection places with GPS coordinates, plants, and insects associated with Zelus findings. The LABs for this study originate from collections that began in August 2014 and ended in December 2019. Images (in .raw file format) illustrate the collection site and context. We registered all data collected in a FileMaker® database (version Pro 12, Claris International Inc, Cupertino, CA, USA).

Table 1. Zelus rewarldii field survey in southern: Italy, Albania, and Spain.

| Locations                                      | Date         | GPS               | Plant Exploitation | Associate Insect                                      |
|-----------------------------------------------|--------------|-------------------|--------------------|------------------------------------------------------|
| “2 Giugno” Public Park, Bari, Italy           | 24 September | 41.10250055° N;   | Quercus ilex L., 1753 | *Nidularia pulvinata* Planckh, 1864; *Kermes vermilio* Planckh, 1864 |
|                                               |              | 16.8747697° E    |                    |                                                      |
| Campus “E. Quagliariello” University of Bari  | 26 September | 41.103763° N;     | Citrus spp., 1753;  | *Aleurocanthus spiniferus* (Quaintance, 1903); *Aleurothrixus floccosus* (Maskell, 1895); |
| Aldo Moro, Italy                              |              | 16.859629° E     | *Pyracantha coccinea* M. Roem., 1847; *Pyrus communis* L., 1753; |
|                                               |              |                   | *Olea europaea* L., 1753; *Saissetia oleae* Olivier, 1791; *Homotoma ficus* (L., 1758) |
|                                               |              |                   | *Ficus carica* L., 1753; |                                                      |
### Table 1: Location and Date of Sampling

| Location                                      | Date       | Latitude          | Longitude         |
|-----------------------------------------------|------------|-------------------|-------------------|
| “A. De Gasperi” Avenue 268, Bari, Italy       | 6 August 2015 | 41.104161° N;      | 16.871717° E      |
| “Papa Pio XII” Avenue 32, Bari, Italy         | 21 August 2015 | 41.103763° N;      | 16.859629° E      |
| “L. De Laurentis” Avenue, Bari, Italy         | 22 September 2015 | 41.093992° N;   | 16.864317° E      |
| S.S. 271, Km 9 Bari-Bitritto, Italy           | 15 April 2016 | 41.066386° N;      | 16.826862° E      |
| “G. Mazzini” Avenue, Valenzano, Italy         | 19 April 2016 | 41.04405° N;       | 16.88717° E       |
| “Roma” Avenue, Copertino, Lecce, Italy        | 18 May 2016  | 40.268763° N;      | 18.050435° E      |
| “G. Toma” street 17, Bari, Italy              | 19 September 2017 | 41.108673° N; | 16.872666° E      |
| Campus “E. Quagliariello” University of Bari Aldo Moro, Italy | 20 September 2017 | 41.103763° N; | 16.859629° E      |
| “Azienda Martucci” farm University of Bari Aldo Moro, Valenzano, Bari, Italy | 22 September 2017 | 41.022620° N; | 16.904295° E      |
| “E. Montale” Street, Copertino, Lecce, Italy  | 03 March 2018 | 40.263238° N;      | 18.056953° E      |
| “Piazza dei Martiri” Gioia del Colle, Bari, Italy | 29 September 2018 | 40.800365° N; | 16.922870° E      |
| “XXV Luglio” Avenue, Lecce, Italy             | 02 October 2018 | 40.353825° N;    | 18.173999° E      |
| “Carrer Portugal” Alicante, Spain             | 15 September 2018 | 38.337504° N; | 0.492043° E       |
| “Demokracia” Street, Tirana, Albania          | 01 March 2019  | 41.3715552° N;    | 19.7776646° E     |
| “A. Jashari” Street, Laknas, Albania          | 04 March 2019  | 41.3687091° N;    | 19.7426623° E     |

### 2.3. Laboratory Work

Our tests aim to understand the LAB predatory attitude versus Palearctic vicariant prey in Italy. These tests compare the use of the referenced *Zelus*-sympatric prey in North America with those vicariant in Italy.

LAB rearing and tests were performed in our laboratory, between 18 and 25 °C and enlightened by side windows, apart from background artificial ring lighting during the Aphrophoridae and honeybees’ tests.

Table 2 summarises rearing arenas and performed tests, revealing the LAB broad behaviour and efficacy versus eleven different prey species of different instars, living either motile or immotile, on their host plants or other backgrounds. Laboratory rearing or testing depended on insect availability for continuous living prey supply.
We used LABs of both sexes to test prey acceptance, apart from the trial on *Bactrocera oleae* Gmelin, 1790, since only predator males were available at the time in the laboratory. In the predation tests, LAB and potential prey had 10 min to interact, if not reported otherwise. When interactions lasted longer than 10 min, tests were left to continue until the interplay between LAB and prey remained.

Table 2. *Zelus* predation test arenas details.

| Prey Species                  | Prey Taxa                        | Cage Area       | Cage Volume       | Prey Density | Notes                                                                 |
|------------------------------|----------------------------------|-----------------|-------------------|--------------|------------------------------------------------------------------------|
| *Macrophomota gladiata*      | (Hemiptera: Psyllidae)           | 9.18 × 7.12 × 8.9 cm | Approx. 580 cm³   | > 0.06 prey/cm³ | Nymphs aggregate on deformed twigs. Adults are vagrant.              |
| *Planococcus citri*          | (Hemiptera: Pseudococcidae)      | 8.55 cm (Ø); 1.42 cm (h) | 81 cm³          | 0.06 prey/cm³ | *Zelus* only preys on moving individuals                               |
| *Planococcus ficus*          | (Hemiptera: Pseudococcidae)      | 8.55 cm (Ø); 1.42 cm (h) | 81 cm³          | 0.06 prey/cm³ | *Zelus* only preys on moving individuals                               |
| *Aleurothrixus floccosus*    | (Hemiptera: Aleyrodidae)         | 9.18 × 7.12 × 8.9 cm | Approx. 580 cm³   | circa 0.6 prey/cm³ | Approx. 50 individuals per leaf                                      |
| *Aleurocanthus spiniferus*   | (Hemiptera: Aleyrodidae)         | 9.18 × 7.12 × 8.9 cm | Approx. 580 cm³   | circa 0.6 prey/cm³ | Approx. 50 individuals per leaf                                      |
| *Latilica sp.*               | (Hemiptera: Issidae)             | 8.55 cm (Ø); 1.42 cm (h) | 81 cm³          | 0.01 prey/cm³ | In mixed prey ensemble                                                |
| *Kermes vermilio*            | (Hemiptera: Kermeidae)           | 9.18 × 7.12 × 8.9 cm | Approx. 580 cm³   | min. 1.2 prey/cm³ | 2–3 adult females and hundreds of crawlers                           |
| *Nidularia pulvinata*        | (Hemiptera: Kermeidae)           | 9.18 × 7.12 × 8.9 cm | Approx. 580 cm³   | min. 1.2 prey/cm³ | 2–3 adult females, hundreds of crawlers                              |
| *Saissetia oleae*            | (Hemiptera: Coccinellidae)       | 9.18 × 7.12 × 8.9 cm | Approx. 580 cm³   | From 0.02 to 0.03 prey/cm³ | 2–3 nymphs underside each olive leaf                                |
| *Harmonia axyridis*          | (Coleoptera: Coccinellidae)      | 8.55 cm (Ø); 1.42 cm (h) | 81 cm³          | 0.01 prey/cm³ | LAB preys the ladybeetle without killing it                           |
| *Apis mellifera*             | (Hymenoptera: Apidae)            | 8.55 cm (Ø); 1.42 cm (h) | 81 cm³          | 0.01 prey/cm³ | Workers                                                               |
| *Philaenus spumarius*        | (Hemiptera: Aphrophoridae)       | 10 × 10 × 5 cm | Approx. 500 cm³   | 0.02 prey/cm³ | As in [11]                                                           |
| *Neophilaenus campestris*    | (Hemiptera: Aphrophoridae)       | 10 × 10 × 5 cm | Approx. 500 cm³   | 0.02 prey/cm³ | As in [11]                                                           |
| *Philaenus spumarius*        | (Hemiptera: Aphrophoridae)       | Falcon         | 60 cm³           | from 0.15 to 0.25 prey/cm³ | In mixed prey ensemble                                                |
| *Neophilaenus campestris*    | (Hemiptera: Aphrophoridae)       | Falcon         | 60 cm³           | from 0.15 to 0.25 prey/cm³ | In mixed prey ensemble                                                |
| *Issidae*                    | (Hemiptera)                      | Falcon         | 60 cm³           | from 0.15 to 0.25 prey/cm³ | In mixed prey ensemble                                                |
| *Miridae*                    | (Hemiptera)                      | Falcon         | 60 cm³           | from 0.15 to 0.25 prey/cm³ | In mixed prey ensemble                                                |
| *Bactrocera oleae*           | (Diptera: Tephritidae)           | 9.18 × 7.12 × 8.9 cm | Approx. 580 cm³   | 0.03 prey/cm³ | LAB preys the olive fly                                               |
2.4. LAB Rearing

*Zelus* were reared continuously in the laboratory in Petri dishes (Figures 2a,b) of various sizes (Table 2). Vented polystyrene rearing flasks also hosted these reduviids, depending on their instar. Crystal-clear methacrylate or high-density polyethylene also provided excellent observation opportunities. Pieces of water-soaked paper or cotton wool maintained the proper environmental RH% (Relative Humidity), and a filter paper disk floor offered an appropriate substrate for the insects. Only Italian *Zelus* was reared in the laboratory. *Zelus* collected abroad were immediately killed and preserved in 80% EtOH (Ethanol).

![Figure 2. (a,b) LAB female reared in a Petri dish on jasmine twigs with meadow spittlebugs available prey.](image)

*Zelus*, paired into a 9 cm Ø Petri dish (Figure 3), later returned to individual rearing plates. Newly born naiades were moved to new containers to give them adequate space. Non-single use rearing arenas were cleaned and sanitised with 70% EtOH every 48 h to reduce the risk of contamination.

![Figure 3. LABs mating in the laboratory.](image)
2.5. LAB Rearing on Drosophila melanogaster, Drosophila suzukii, and Megaselia scalaris

_Drosophila melanogaster_ L., 1758, was grown in 4 L plexiglass flasks on modified [11] meridic artificial substrate [22]. _Drosophila suzukii_ (Matsumura, 1931) was grown on ripened and overripened fruits available in the season. _Drosophila suzukii_ is an alien invasive pest in Italian vineyards [23,24], which has recently acclimatised to Apulia [25]. _Megaselia scalaris_ (Loew, 1866) was reared on espresso coffee wet waste capsules placed in an opaque wastebasket.

2.6. LAB Rearing on Macrohomotoma gladiata

Fresh watered _Ficus_ sp. twigs in polystyrene-vented rearing flasks (Table 2) harbouring active _M. gladiata_ adults and nymphs hosted single wild adult _Zelus_. Psylla was paired and bred in flasks with _Zelus_. The twigs maintained proper moisture and supported both predator and prey. The psyllid _M. gladiata_ is an alien invasive pest in the EPPO region [26,27].

2.7. LAB Test on Pseudococcidae

_Planococcus citri_ (Risso, 1813) and _Planococcus ficus_ (Signoret, 1875) were reared on butternut squashes (_Cucurbita moschata_ Duchesne ex Poir., 1818) [28,29], in CI-HEAM-IAMB facilities or collected on grapes. We placed small mealybugs groups into Petri dishes (Table 2).

2.8. LAB Test on Aleyrodidae

Fresh _Citrus_ sp. twigs (40 cm long and with 15–20 leaves each) infested by _Aleurocanthus spiniferus_ (Quaintance, 1903) and _Aleurothrixus floccosus_ (Maskell, 1896)—underside the leaves—were placed in polystyrene-vented rearing flasks (Table 2).

2.9. LAB Test on Coccomorpha

Fresh _Q. ilex_ twigs (30 cm long, with 20–30 leaves each) infested by _Kermes vermilio_ Planchon, 1864, and _Nidularia pulvinata_ (Planchon, 1864) served to test LAB predation. _O. europaea_ twigs (20 cm long, with 30–35 leaves each) infested by _Saissetia oleae_ (Olivier, 1791) hosted LAB predation tests on Coccidae. These tests were performed in polystyrene-vented rearing flasks (Table 2).

2.10. LAB Test on Xylella vectors, Miridae and Issidae

Aphrophoridae, Miridae and Issidae were captured using sweeping conifers by nets in 5 min rounds. A large funnel was used to transfer hemipteran into a 5 L PET-vented plastic tank. Avoiding the use of aspirators prevented a de-pressuring shock and the consequent unpairing of the prey. The sweeping also gathered twigs of _Cupressus sempervirens_ L., 1753, _Cupressus arizonica_ Greene, 1882, and _Thuja occidentalis_ L., 1753 food plants. These were kept for moisture and fed to the prey. Each test used only 15, fully active Hemiptera adults, to ensure no attempts to escape from the tank.

2.11. LAB Test on Harmonia axyridis

Twenty-three _Harmonia axyridis_ (Pallas, 1773) were collected on _Citrus_ sp. infested by whiteflies (_A. spiniferus_ and _A. floccosus_) in the University of Bari Aldo Moro Campus tested with LAB. Each ladybird interplayed in a Petri dish (Table 2) with one LAB.

2.12. LAB Test on Bactrocera oleae, the Olive Fly

Eleven LAB adults preyed on 10–12 _B. oleae_ during a one-day extended test, intermingled with a day of starvation. The container for the test was a vented parallelepiped polystyrene rearing flask (Table 2). The olive fly puparia were from an oil mill near Molfetta (41.205651° N; 16.573373° E). Adult olive flies escaped from puparia in plexiglass-
vented boxes (Table 2) with a plenty food supply [30]. To avoid using teneral adults, the flies hardened in the boxes for a minimum of one day after the escaping and before the tests. We recorded LAB feeding time on each B. oleae as we did for the Xylella vectors tests.

2.13. LAB Test on Apis mellifera

We investigated the ability of Z. renardii to prey on Apis mellifera L., 1758 (Hymenoptera: Apidae—AM). We placed both insects in Petri dishes (Table 2, 81 cm³) on shading support over a LED (4000 K°) ring lamp for 10 min to interact. We used 19 LAB individuals (9 ♀ and 10 ♂) and five mid-aged honeybee workers. Each AM replayed randomly ca. four times. Insect behaviour was recorded (.raw image or full HD movie format) with a digital camera (Olympus® Pen camera, OM Digital Solutions Corporation, Hamburg, Germany). If an interaction lasted more than 10 min, we continued to record until the interplay ended. Scrutinising the interactions resulted in a series of paths of both insects, drawn to follow the tracks taken by LABs and AMs. Path drawings were processed using Adobe Illustrator® (Adobe Inc., San Jose, CA, USA).

2.14. LAB and Humans

We reproduced LAB biting events as reported on humans. Ten volunteers placed LABs on the naked skin of the back of their left hand, pushing them progressively and firmly to elicit LAB biting. We recorded the LAB action and marked the piercing point, following its evolution for a week. None of the volunteers had a known history of allergy requiring special care. All LAB died during the tests based on the pressure they had endured.

2.15. LAB’s Tests Interpretation Issues

The LAB predation tests here presented did not consider any functional response [31,32]. The tests only supported the data of LAB Nearctic prey during more than a century of studies on various European vicariant prey.

The reduviids’ abdominal fold (Figures 4a–c) and defecations indicate feeding after each attack. The status of the abdominal fold allows recognising a starved or engorged individual. Usually, egg laying follows the engorgement of a LAB female (Figure 5).

Figure 4. (a) starved LAB female displays contracted abdominal fold at the feeding start; (b) LAB female with semi-contracted abdominal fold after egg laying, dark-yellow spots are faeces; (c) LAB female shows distended abdominal fold after preying.
Figure 5. LAB female during egg laying.

Post-predation egg laying and hatching of LAB after tests indicate if that particular prey is suitable for predator reproduction. Results clearly state if eggs are missed.

3. Results and Discussion

3.1. References Retrieval

Literature searches yielded 1235 records. The analysis discarded 191 duplicates plus 880 untraceable documents. Furthermore, seven new articles originated from nested reference scrutiny. Our literature search yielded 171 references for the current study. These references represent 16.27% of the entries retrieved and 13.85% of the total titles (including duplicates) found in the databases.

The references [33–124] gathered information on LAB bionomics and prey preferences (Table S1), ordered per taxon, types of approach, and qualitative or quantitative experience. The literature reports several observations of LAB feeding on various prey, mainly from the Nearctic region. Only a few reports exist for LAB prey from other biogeographical areas.

The literature of LAB prey preferences showed that LAB uses 88 prey species belonging to nine orders. References show ca. 48.9% of the total prey species belong to Hemiptera. Other taxa references—Blattodea and Orthoptera 1.1%, Thysanoptera 4.6%, Neuroptera 5.7%, Lepidoptera 15.9%, Coleoptera 12.5%, Diptera 4.6% and Hymenoptera 5.7%, respectively. The analysis of Hemiptera species references in Figure 6 shows a wide array of prey, with a 20.9% maximum score for Aphididae.
Beneficial insects as *Chrysoperla carnea* (Stephens, 1836), *Adalia bipunctata* (L., 1758) and *A. mellifera* only represent the 2.4%, 1.2% and 1.2%, respectively, of LAB’s predation events under natural conditions, as shown in Table S1. LAB can predate more on them in constricted environments (i.e., cages, boxes, etc.).

Therefore, LAB appears much more stenophagous [19] on Hemiptera than a polyphagous predator. *Zelus longipes* (L., 1767), just as *Z. renardii*, selects prey based on their mobility and size [125].

Literature metadata (Supplementary Material: Table S1) shows that field observations represent 59.5% of total references, the remaining 40.5% being laboratory observations. Reports about LAB prey preferences are qualitative (51.8%) and quantitative (48.2%) in similar percentages. Field experiments report much regarding LAB preference for a certain prey and represent non-quantitative observations, not valid for pest biocontrol forecast and impact evaluation on beneficial arthropods. Furthermore, they do not discuss intra-guilt predation.

*Zelus* is amongst the largest genera of the Reduviidae [126–129], with ca. 70 described species [33,126,130]. Kolenati [131] described *Zelus renardii* in 1856, and Weirauch [130] accepted the “Leafhopper Assassin Bug” (LAB) as its trivial name. Henry [132] and Zhang [126] revised the genus species placing *Zelus cervicalis* Stål, 1872 as a sister species of *Zelus renardii* but accepting *Zelus peregrinus* Kirkaldy, 1902, *Zelus laevicollis* Champion, 1898, and *Diplodus renardii* Uhler, 1894, as synonyms.
Zelus renardii is native of North and Central America [33,42,126,129,130,133–137]. The reduviid widely spread throughout the American continent [42,123,138–144], Asia and Oceania [143,145–149]. The bug also entered Hawaii [150], and Zimmerman [71] considers Zelus a large, mostly tropical American genus.

Several authors [33,151] have reported LAB from mainland Greece, as Simov [152] confirmed. Later, van der Heyden [153] found Zelus in Crete. During the same period, Zelus entered Spain [133,134], Italy [135,154–156], Albania [127], France [157] and Portugal [158]. However, recently, LAB has entered Germany [159], Denmark and the UK via grape transport from Italy and Greece [160]. Today, LAB is considered well established in Europe [136] as well as in Italy [137] and the Iberian Peninsula [161–163].

LAB is an alien, acclimated, spreading species in many Mediterranean areas adapted to urban and peri-urban ecosystems [164,165].

Miller [17] considers Reduviidae generalist predators that prey opportunistically on various arthropods, including beneficial insects.

Qualitatively, LAB is a predator of different pests and beneficial insects (Table S1) in the field and laboratory conditions. Quantitatively, its favourite preys belong to the Hemiptera [166] and the Cicadellidae [58], Aphidiidae [12,13,54] and Psyllidae [1]. Ambrose [16] considers LAB as a “generalist” when introduced and a “specialist” insect pest predator in its habitat. The difficulty is to place this predator in a sharp category, ranging from monophagous specialists to ultra-specialists. However, assigning a particular non-specialised predator to a specific behavioural class (polyphagous, stenophagous, oligophagous) [19] is not immediate due to our limited knowledge of the species diet in nature [12–15].

LAB is almost an exclusive carnivore [167], differing from many stenophagous and generalist predators that also eat plants and even pests. Mixed-diet insects might reasonably be called generalist omnivores [19]. LAB’s prey choice varies according to ecological conditions such as plant habitat, composition, and abundance of prey species. In the absence of prey, Z. renardii naiades could supplement their diet with vegetable-derived pabula to survive for short periods [14,168]. Under severe starvation, LAB can turn to cannibalism [1,12,58,82,106], eventually leading to the self-control of the population. LAB is highly adaptive, dispersive, and able to colonise new areas. This may generate density of 50,000–75,000 individuals/ha [105].

LAB prefers mobile prey [58] and does not play an important biological control agent role in cotton fields [169]. LAB presence in orchards and crops depends on prey abundance or prey diversity [105] and mobility. LAB adults show a functional response to prey abundance [100], size [67,106] and the presence of honeydew-producing Hemiptera. The LAB predation strategy depends mainly on prey size [16,130,170,171]. Zelus prefers to ambush small prey but to stalk larger ones [105]. A sticky secretion on the anterior tibiae has a decisive role in predation success [137,139,140,170–174]. This secretion of the forelegs allows sticking the prey on the anterior legs minimising attack failures. Generally, Zelus is quite active and voracious with a low attack failure rate. Furthermore, the salivary glands’ enzymes allow ingesting large amounts of prey body mass [175].

LAB lays eggs in batches, each with 12 to 62 units [58]. Each female can lay up to 270 eggs [138] in 2–3 sets during its life [43,112], with a fertility rate ranging from 18 to 64% [43]. The eggs are sub-cylindrical, brown [58,71,122,138,172] and topped by a whitish operculum [58]. Moreover, LAB covers egg masses with a sticky secretion [58,138]. Usually, the reduviid places the egg batches on the upper side of leaves or various substrates. Post-embryonic development consists of five juvenile instars [58,138]. Adults exhibit sexual dimorphism, with bigger females (♀♀ are about 14 mm long and ♂♂ ca. 11 mm) [58,176].

LAB is a predator with a preference for disturbed environments [33,105]. It is a synanthropic [42] insect occurring in urban and peri-urban environments [42,43,114,130,137,161], where it has a greater chance of encountering various prey species.
Human activities facilitate *Z. renardii* passive dispersion, facilitating entrance and exploitation of new countries [130,141,143,145]. The positive correlation between urban environments and reduviid abundance also confirms human-driven LAB passive dispersion.

Force [177] considers r-strategist predators such as *Zelus* less numerically dominant in mature ecosystems and regularly found on a broad geographical or ecological range. Furthermore, r-predators tend to be abundant in perturbed habitats (e.g., agroecosystem or urban areas). They also have a high demographic growth rate being relatively weak competitors. The reduviid life cycle completes in about two months [43,71].

The numerical and functional responses to prey density do not govern a dynamic relationship between the LAB and target prey. Moreover, predators choose specific targets to supplement their amino acid requirements [178].

3.2. Field Observations and Tests

LAB synecology and biology feature displayed in Table 3. It is easy to spot LAB hunting in urban and peri-urban areas frequenting hemipteran-infested plants. We collected a total of 126 LABs (Table 3).

**Table 3.** *Zelus renardii* presence in plant and associated honeydew-producing Hemipteran pests. Data refer to Table 1. Abbreviations: N1–N5 = juvenile instars; Ad = adult.

| Host Plant          | Associated Hemipteran | LAB Instars |
|---------------------|-----------------------|-------------|
|                     | Egg                   | N1 | N2 | N3 | N4 | N5 | Ad |
| Quercus ilex        | Nidularia pulvinata    | 4  |    |    |    |    |    |
| Quercus ilex        | Kermes vermilio       | 1  |    |    |    |    |    |
| Olea europaea       | Saissetia oleae       | 1  |    |    |    |    |    |
| Olea europaea       | Saissetia coffeae     | 2  |    |    |    |    |    |
| Ficus microcarpa    | Macrohomoloma gladiata| 1  | 1  |    |    |    |    |
| Ficus carica        | Homotoma ficus        | 2  | 1  | 4  |    |    |    |
| Ailanthus altissima | Aleurocanthus spiniferus| 7 | 1  | 2  | 1  |    |    |
| Pyracantha coccinea | Aleurocanthus spiniferus| 1 | 1  | 2  | 5  |    |    |
| Pyrus communis      | Aleurocanthus spiniferus| 14|     | |    |    |    |
| Citrus CV           | Aleurocanthus spiniferus| 1 | 2  | 3  | 18 |    |    |
| Citrus CV           | Aleurothrixus floccosus| 12| 1  | 3  | 4  |    |    |
| Aloysia citriodora  | Aphis spp.            |    | 1  |    |    |    |    |
| Capsicum annuum     | Aphis spp.            |    | 2  |    |    |    |    |
| Chamaerops humilis  | Ommatissus binotatus  | 6  | 1  | 2  | 5  |    |    |
| Laurus nobilis      | Trioza alacris        | 11 | 1  |    |    | 1  |    |

LAB collection of wild adults outnumbers the number of eggs and juveniles on plants (Table 3). Assuming a population in equilibrium, the number of eggs and juveniles should be higher than adults [179,180]. The adult population should result in fewer adults than the corresponding eggs and juveniles, contrarily as reported in Table 3. The total number of adults found (64 LABs) is larger than the sum of the eggs, and juvenile stages found (33 and 29, respectively). Adults’ collection is more accessible than pre-imago instars because the individuals are prominent and easier to spot than smaller, slender juveniles. Adults prefer large prey, and require more *pabula* than juveniles. The greater mobility of adults also makes them more visible in the field.

The hemipteran’s honeydew could lure LAB to the plants infested with available prey, causing LAB egg laying and the possible presence of their pre-imagoes. Multiple observations of *Zelus* preying on wandering among *T. alacris, O. binotatus, A. spiniferus, S. coffeae* or *S. oleae* suggest this honeydew luring effect. Fall surveys on the same plants listed in Table 3 did not result in LAB findings. Perhaps LAB adults had moved to overwinter
Further comparative observations on neighbouring clean plants support the hypothesis that LAB is associated with hemipteran honeydews or faeces.

Honeydew-like fluids, or some of the attractive semiochemicals within, could influence Zelus behaviour. Honeydew could push the predator to aggregate and persist in a specific place for pest control. The Hemiptera honeydew likely contains volatile organic compounds (VOCs) that may act as kairomones attracting the predator. Other VOCs could originate from honeydew or artificial surrogates or associated microorganisms either drawing antagonists [182–185] or attracting/repelling pests [186]. These VOCs may participate in behavioural manipulation actions within an IPM strategy to enforce any non-synthetic chemical control action arsenal for bio-management of Xf vectors.

LAB mating lasts about 37 min (Table S2) under laboratory conditions (T = 25 ± 3 °C and RH = 80 ± 5%). Females reject the partner shaking the abdomen [58]. Starved females may try to prey on the approaching male or kill it after the mating. Cannibalism is not a novelty in LAB female rearing, however, male killing is not regular and does not have the sense to replace missing preys.

Mbata [112] and Barrera [43] suggest that each LAB female laid 2–3 batches giving an average of 33 eggs with a prolificacy ranging from 18 to 64% [43]. In our rearing, females laid a total of 1112 eggs; females laid an average of three batches averaging 27 eggs (Table S2) [58,138] with a prolificacy of about 80%, with a higher hatching rate (Figure 7) and many new-borns than reported. Moreover, females are polyandrous, and El-Tom [58] considers repeated mating induces more eggs than single-mating. Embryonic development of our LAB lasts 17.4 ± 5.4 days, unlike Swezey [138] and El-Tom [58] that reported LAB embryonic development of about 6–10 days at 25 °C. Our hatching period data is the average value for one-year-long LAB breeding experiments. Swezey [138] and El-Tom [58] report a shorter May–August breeding period. Differences in hatching time may rise from photoperiod, food available or female age. There is a concordance between our breeding data and observations of one-century-long literature.

Figure 7. Hatching percentage of thirty egg batches laid after LABs mating, detailed in Table S2. Abbreviations: Eb = egg-batch.

3.3. LAB Rearing on Drosophila melanogaster, Drosophila suzukii and Megaselia scalaris

Zelus can be mass reared successfully with a continuous supply of D. suzukii, D. melanogaster and M. scalaris. Interestingly, D. suzukii is now another phytophagous species on olive fruits [187]. Therefore, models and joint strategies for bio-management of Xylella vectors (key pest) and secondary olive pests, as D. suzukii, should consider this LAB attitude.

The LAB’s juveniles fed on maggots and adults (Figure 8a,b) of both the gnats during the rearing, showing a normal post-embryonic development until adults. Adult reduviids were active, and females laid regularly fertile egg batches that hatched as expected on almost all available surfaces. The egg laying proved that these dipterans are suitable prey for LABs mass rearing.
3.4. LAB Rearing on Macrohomotoma gladiata

In rearing, each reproductive LAB uses 25–30 M. gladiata adults per day plus an unknown number of nymphs (Figure 9a,b). Macrohomotoma gladiata juveniles are uncountable because sheltered among fig leaves.

3.5. LAB Tests on Pseudococcidae

Zelus, in general, ignores immotile prey such as Pseudococcidae. Few predations occur when P. ficus and P. citri move, provoking the immediate reduviid action that spots and pierces them (Figure 10). In this case, feeding lasts 1–2 min. LAB could feed on Pseudococcidae, but it does not lay eggs.
Figure 10. LAB female feeding on Planococcus citri adult.

3.6. LAB Tests on Aleyrodidae

LAB females survive 7–10 days on mandarin twigs harbouring A. spiniferus and A. floccosus mixed infestations without laying eggs. Aleurocanthus spiniferus is an alien invasive Aleyrodidae, representing the new key pest for Citrus spp. orchards in Apulia [188,189]. Few whitefly adults were occasionally preyed upon [51]. Any whiteflies (ca. four individuals for each LAB) died because glued to the predator’s sticky forelegs [174,190], but LAB neither fed on those bodies nor laid eggs.

3.7. LAB Tests on Coccomorpha

There is no evidence of LAB predation either on an adult or on crawlers (naiades) of K. vermilio, N. pulvinata and S. oleae. Some crawlers died, stuck to the predator’s glutinous forelegs. No egg laying occurred.

3.8. LAB Tests on Xylella vectors, Issidae and Miridae

LAB preys on a plethora of different phytophagous Hemiptera belonging to Aphrophoridae, Issidae, and Miridae. Observations showed that LAB captures its target first, sticking it with the forelegs [174,190]. The insertion of the stylets through the prey intersegmental membranes follows. The saliva injection kills the prey, possibly because the action of proteolytic enzymes (proteinase and endopeptidase) predigest the prey [102]. Afterwards, the predator sucks the lysate obtained from the prey body [102].

Figures 11 and 12 show the feeding time for the 21 LAB (eight females and 13 males) predations experiment on mixed Aphrophoridae, Miridae and Issidae with a maximum of 15 prey per LAB.

Figure 11. Z. renardii average feeding time. Each bar represents the average time that LABs took to feed on each prey.
Zelus preys by ambushing or stalking its prey [105]. LAB predation that occurs within a 60 cm³ Falcon flask happens primarily due to accidental encounters between moving predator and prey rather than LAB aggressions. Indeed, the high prey availability creates a great chance of casual prey-predator encounters that prevent LAB from executing its conventional predatory strategies because there is always an available prey around. What we see in Falcon tubes are LABs waiting for the passage of prey to attack and kill it. Prey was so frequent that the LAB stopped feeding on the sixth prey onwards. LABs continued to kill the next prey but did not feed on them.

In Falcon tube experiments, the attack time diminished by the frequencies of the encounters with the prey, and LAB devoted remaining time to feeding only. Thus, the feeding time depends mainly on the prey size and LAB starvation [66,106,125,130,170,171]. LAB takes on average 33 min 5 s to feed on each prey. Overall, predator feeding time decreases with the increasing number of predations. The longer feeding time (73 min 17 s) corresponds to the first prey, and the shorter (5 min 8 s) coincides with the last one.

The trend line shows two time-plateaux splits by an abrupt drop of feeding time, corresponding to the prey from the 7th to the 9th. First plateau—prey 1st to 6th (61 min 19 s)—corresponds to a real feeding time, but the second plateau—prey 10th to 15th (08 min 57 s)—accounts for just killing and carcass abandonment, with any or negligible feeding on prey. The behaviour strongly suggests that engorgement rules the LAB, which does not feed even if it still follows its killing instinct.

LABs show a high predatory aptitude on Xylella vectors, killing almost all available Aphrophoridae, Issidae and Miridae. Indeed, LAB laid eggs batches after preying on Aphrophoridae, Issidae and Miridae.

During tests, LAB reused prey carcasses. In the Falcon tubes, LAB can easily be in touch with prey it killed previously. Often the predator drives the stylets in that dead carcass as it does with just killed insects. The behaviour may be relevant in restricted spaces where carcasses are still available. Carcass feeding is possibly negligible in the wild because dead prey abandonment provokes their dispersal in the environment. In small arenas with high prey density, overrepresented carcass feeding occurs. The observation seems a behaviour not previously reported in the literature. The use of such carcasses may help avoid cannibalism in overcrowded LAB mass rearing, eventually.

Differences exist among females and males prey use (on average, ♀ 44 min 39 s vs. ♂ 33 min 44 s). Apart from sex-related prey exploitation, wild LAB could host not sex-related and non-lethal but weakening viruses, bacteria, apicomplexa, and fungi (yet to be found). LAB collected in the wild has unknown age or sanitary status.
3.9. LAB Tests on Harmonia axyridis

Twenty-three *Harmonia axyridis* underwent attacks in Petri dishes by five LABs (two ♀ and three ♂) that stalked them. Despite the immediate beetle immobilisation after apparent stylet insertion (Figure 13), victims soon recovered from the aggression by walking around; this differs from previous reports [114]. The same LAB also re-attack surviving *H. axyridis* during a subsequent encounter in the Petri dish. We did not test if ladybirds were castrated by the attacks or suffered other significant damages. After this episode, LAB did not release eggs, and the abdominal ventral fold stretched moderately.

![Figure 13. LAB male feeding on Harmonia axyridis.](image)

3.10. LAB Tests on Bactrocera oleae (BO, the Olive Fly)

LABs attack fruit flies, *Bactrocera cucurbitae* (Coquillett, 1899), in the field [65,113] and thrive on *Ceratitis capitata* (Wiedemann, 1824) and *Anastrepha ludens* (Loew, 1873) [43]. In laboratory tests, LAB males also attack *B. oleae* (Figure 14), killing 10–12 individuals exposed in the arena (Table 2, 580 cm²) within 24 h. Feeding time was about 60 min for the first prey but shortened to about 18 min for the 12th. The prey dynamics are consistent with those observed in the Aphrophoridae, Issidae and Miridae tests. Once engorged, LAB reduced the feeding time to the last prey. The remaining BOs disturbed LAB feeding by flying and banging over the reduviid. LABs replied by leaving the feeding prey and killing the BO that irritate them. In total, only five BOs managed to escape from the LAB’s attack.

![Figure 14. LAB male feeding on Bactrocera oleae adult.](image)
Bactrocera oleae was the key pest of olive trees in southern Italy until evidence of transmission of Xf via Aphrophoridae was formed [4]. From an IPM perspective, predation of BO would allow to manage it as a pest and sustain LAB on the olive orchards as alternative prey to Xylella vectors. Indeed, the scarcity or absence of prey would induce the LAB to abandon the olive orchards in search of a new target.

Average feeding times per ambushing predator shows similar lengths, possibly because this test included males only, instead of both males and females as for Aphrophoridae prey (Figure 15). Abdominal ventral fold stretches as LAB feeds on the first prey. LAB takes on average 38 min 36 s to feed on each BO (Figure 16). Males only participated in the test because of vagrants available in winter.

![Figure 15. Zelus renardii feeding time in minutes recorded in 580 cm³ arena each provided 10–12 prey, details in Table S4. Each bar represents the average time that Z. renardii took to feed on each prey item.](image)

![Figure 16. Zelus renardii feeding time per prey in minutes recorded in 580 cm³ arena per each of the 11 predators, details in Table S4. Each bar represents the average feeding time per 10–12 prey items.](image)

3.11. LAB Tests on Apis mellifera (AM)

One of the major concerns against LAB use for inundative releases in the field was the claim about reduviid predation on honeybees. Our experiments with LAB and AM placed together in Petri dishes (Table S5) show five behavioural patterns: (I) reciprocal ignorance, wandering around in the Petri dish; (II) staying almost immotile near or on the release spot; (III) AM walks or flutters versus the escaping LAB; (IV) AM accidentally collide with LAB eliciting its aggressive posture; (V) AM glues on LAB sticking legs that attacks and kills the honeybee defending itself (Figure 17).
LAB & AM stand from 02:17 to 02:47

LAB Ambushes from 05:38 to 06:08

05:09 LAB faces AM, projects the rostrum, touches AM with the tip of the last antennomere, and withdraws

LAB Ambushes
Most (73.7%) LAB–AM interactions (Table S5) belong to type I behaviour: reciprocal overlooking with both actors wandering alone in the plate.

In type II behaviour (Table S5), LAB and AM stay motionless, staring at each other at the release point.

Walking across the Petri dish (Table S5), LAB cautiously touches AM with its antennae and then abruptly withdraws in fear as type III behaviour.

In type IV behaviour (defence), AM rushed through the Petri dish, touching or colliding with LAB (Table S5). LAB raises its anterior legs distending the rostrum in a defensive position. On collision events, LAB may become stuck on AM by the anterior gluey legs. The reduviid moves backwards to break off the AM, stressing the anterior legs up, releasing them from AM seta. This interaction ended without AM pierced.

The test reveals that the 5% only of the forced interaction into a restricted arena led to one attack due to a defensive LAB need (Figure 18). Behaviours following the death of
the honeybee, stylet insertion and use of the carcass are comparable with those of the other prey/carcasses.

Figure 18. LAB female feeding on the single killed AM worker.

The only predation resulted because of the sticking of AM on the LAB forelegs. Based on these observations, we suggest that AM is not among LAB regular prey.

Speed analysis (Figures 19 and 20) of LAB–AM interactions can shed light on LAB predatory behaviour. LAB has shown ambushing capabilities with several preys. Therefore, the lack of LAB attacks of static AM strongly suggests a non-LAB prey status for this beneficial insect.

Figure 19. Apis mellifera speed in 9 cm Petri dish arena (details in Table S5).

Figure 20. Zelus renardii speed in 9 cm Petri dish arena (details in Table S5).
In the AM–LAB interactions, AM moves at an average speed of 3 mm/s (Table S5 and Figure 19) but often—68% of assays—it stays at the release spot. Little or no AMs motility could be related to the onset of starvation, which drives them to die 24–48 h after [191].

LABs show a faster average speed (8 mm/s) and mobility (578 mm) than AM during the interactions (Table S5 and Figure 20). Sex-related differences exist in *Z. renardii*, with females that are on average more mobile (943 mm) and faster (9 mm/s) than males. LAB females increased mobility could be related to a significant need for resources to perform vital functions, i.e., egg laying [192,193].

### 3.12. LAB and Humans

LABs bite humans occasionally but only when menaced [114,194,195]. LABs pierced the skin of eight out of ten volunteers. The piercings originate minor skin rashes a few mm in diameter (Figure 21). The pain is almost negligible and does not persist much after the wounding episode. LAB bites on humans are not frequent, and the consequences are similar to European *Polistes* spp. wasp stings on our experience. The day after the bite, the pain usually disappears, and only one weal was present with a culminant biting point and a 1–3 mm flat and non-infected scab. None of the volunteers reported any concerning clinical effects. The LABs involved in the tests died out from the interaction on the volunteers’ skin.

Reduviids produce a wide variety of toxins in their saliva that are possibly involved in skin reactions to piercing. The effects of the saliva toxins vary among vertebrates [196]. LAB bites may raise concern because some Reduviidae are well-known vectors of Chagas disease [197,198], however, this is not the case with LAB since it is not a hematophagous insect. Piercings are rare also because LAB bites for defence only, as reported for other Reduviidae [194,195]. The only possible risk could therefore be associated with allergic reactions in hypersensitive humans.

### 4. Conclusions

LAB is a Nearctic reduviid that entered the Mediterranean as an alien non-invasive species. This reduviid also acclimated in several circum-Mediterranean countries, roaming mainly in urban and peri-urban areas. The coexistence with humans seems more harmful for insects than humans since LAB provokes entomophobia reactions that often lead humans to kill it.

Gross opinions consider LAB a generalist predator, but evidence suggests that it is primarily stenophagous on Hemiptera. LAB chooses its prey according to their habitats, including the prey host plant, abundance, size and mobility of the species it encounters.
LAB was considered capable of disrupting natural biocontrol by intraguild predation. Still, this adverse side effect is probably due to oversimplified experiments in caging a restricted number of species and individuals in small environments.

*Zelus renardii* often seeks honeydew-contaminated plants because they may host potential prey. Honeydew could lure the reduviid on Hemiptera-infested plants provoking egg laying and subsequent LABs juvenile presence, eventually. Honeydew may aggregate the predator and persist in the place we need for pest control, consequently. Hopefully, future studies will isolate attractive semiochemicals for the manipulation of *Z. renardii* behaviour.

Nevertheless, not all honeydew-producing insects are prey for the *Z. renardii*, showing no interest in immotile instars of hemipteran. Tests confirm that LAB does not feed on Aleyrodidae, juveniles and immotile Pseudococcidae, Kermesidae, and Coccididae. LAB is an insectivore sometimes feeding on honeydews or plant secretions in prey absence, but without growing or laying eggs. The reduviid is not a plant pest or phytophagous.

All the LAB’s instars are cannibalistic in conditions of severe starvation that quickly occur in dense breeding. Adults can attack and kill naiades and nymphs, however, females may also kill males approaching for mating.

The distension of the reduviid ventral abdominal fold during feeding and the subsequent regular egg laying strongly suggest prey availability. Moreover, a prey that guarantees food enough for egg laying in tests may be functional for the predator, either in nature or the field.

Quantitative observations on *Z. renardii* can help us develop this predator as a biocontrol agent. LAB predation on *A. mellifera* and *H. axyridis* are rare events in nature and are also exceptional under laboratory conditions.

*Zelus renardii* can breed continuously with a constant supply of several living prey, namely *D. melanogaster*, *D. suzukii*, *M. scalaris*, *M. gladiata*, Aphrophoridae, Issidae, and Miridae. In rearing conditions, the predator reuses the carcasses of already killed prey.

Our tests confirm some Palearctic pests’ availability as prey, which is vicariant for the corresponding Nearctic species that have been well known for over a century of studies.

LAB displays different aggressiveness amongst the prey, behaving as stenophagous. LAB prefers prey able to meet its developmental and reproductive requirements. Some LAB attacks were driven by caging conditions as a defence reaction (i.e., honeybees). The tests suggest that *M. gladiata*, *P. spumarius*, *N. campestris*, *D. melanogaster*, *D. suzukii*, and *M. scalaris* are available pest prey for *Z. renardii*. Field experiments will further demonstrate the role of LAB as the olive fly (*B. oleae*) biocontrol agent. *D. melanogaster*, *D. suzukii*, and *M. scalaris* may help in *Z. renardii* mass or continuous rearing since those prey can be mass reared on artificial diets. *A. mellifera* does not appear as a prey for *Z. renardii*.

Experiments suggest that LAB is a biocontrol agent of some economically relevant olive trees pests. The LAB features analysed in this work make it a good candidate for inundative biocontrol programmes to manage *Xylella fastidiosa pauca* ST53 current pandemics, reducing the incidence of Xf new infections and transmissions. LAB can integrate olive IPM to mitigate damage by other olive-related pests, such as *B. oleae*.

**Supplementary Materials**: The following are available online at www.mdpi.com/article/10.3390/insects13020158/s1, Table S1: *Zelus renardii* prey species preferences with order and family, the corresponding reference with the types of approach and experiments. Table S2: Raw data regarding *Ze‌lus renardii* mating. The data consider: “Date” of mating; “Mating time (min)” is the time in minutes spent for each couple to complete the mating; “Days After Mating (d)” represents the days spent between mating and egg laying; “Eggs Laid” represents the total number of eggs laid; “Eggs Hatched” shows the total numbers of eggs hatched; “Embryonic Development (d)” shows the days spent between eggs laying and eggs hatching. C = couple. Data graphed Figure 7. Table S3: Raw data regarding *Zelus renardii* preying Aphrophoridae, Miridae and Issidae assemblage. The data consider: “Date” of predation; “Place” considers the locations where trials occurred; “N” shows the progressive number used to identify the predator; “M-F” reports the sexing of predators; “Prey 01–
15” reports the progressive number of prey; “Time Sum” accumulates the minutes’ employees by each predator to feed all prey; “Prey n” reports the total numbers of prey preyed. M = Male; F = Female and Dept. = Laboratory of Department. Data graphed Figures 11 and 12. Table S4: Raw data regarding Zelus renardii preying Bactrocera oleae. The data consider: “Date” of predation; “Place” considers the trials locations; “N” shows the progressive number used to identify the predator; “SEX” reports the sexing of predators; “P 01–12” reports the progressive number of B. oleae tested; “Time Sum” accumulates the minutes’ employees by each predator to feed all prey; “Prey n” reports the total numbers of prey preyed. M = Male; F = Female and Dept. = Laboratory of Department. Data graphed Figures 15 and 16. Table S5: Raw data regarding Zelus renardii and Apis mellifera interaction. The “N” shows the progressive number used to identify the interaction between Z. renardii and A. mellifera; “Interactions Sec” reports the time in seconds of each interaction; “AM track (mm)” shows the distance travelled by A. mellifera in the arena expressed in millimetres; “ZR track (mm)” shows the distance travelled by Z. renardii in the arena noted in millimetres; “AM mm/sec” reports the average speed of the A. mellifera during interaction with the predator; “AM mm/sec” reports the average speed of the Z. renardii during interaction with the pollinator; “Interaction” notes the behaviour of Z. renardii and A. mellifera during trials. Data graphed Figures 19 and 20. Zelus-Apis Movie: The movie shows interaction n.19 (Figure 17) between Z. renardii and A. mellifera. Initially, A. mellifera and Z. renardii stick on Zelus anterior legs. Later, the fluttering honeybee accidentally bumps Z. renardii, eliciting its defence biting reaction that will kill the honeybee. Later, the Zelus feed on dead honeybees (Figure 18) as on other dead insects.

Author Contributions: Conceptualisation, U.P., F.P. and F.G.; validation, N.L., U.P. and V.S.; formal analysis, N.L. and U.P.; investigation, U.P., F.P. and F.G.; resources, U.P. and F.G.; data curation, U.P., F.P. and F.G.; writing—original draft preparation, U.P. and F.G.; writing—review and editing, U.P., S.B.-B., F.P. and F.G.; data visualisation, U.P. and F.G.; supervision, F.P. and F.G.; project administration, F.P.; funding acquisition, F.P. All authors have read and agreed to the published version of the manuscript.

Funding: The present work has received funding from the European Union’s Horizon 2020 research and innovation program under Grant Agreements No. 635646—POnTE “Pest Organisms Threatening Europe” and No. 727987—XF-ACTORS “Xylella Fastidiosa Active Containment Through a multidisciplinary-Oriented Research Strategy”. The EU Funding Agency is not responsible for any use that may be made of the information it contains. This research was also supported by CURE-Xf, an EU-funded project coordinated by CIHEAM Bari (H2020-Marie Sklodowska-Curie Actions—Research and Innovation Staff Exchange. Reference number: 694353).

Data Availability Statement: The authors can share the references used to write this contribution.

Acknowledgments: Authors thank Nuray Baser, CIHEAM Bari, for her suggestions and Pseudococcidae loan, Rocco Addante, University of Bari Aldo Moro, and Luis Vicente Lopez-Llorca, University of Alicante, for their reading the draft and comments.

Conflicts of Interest: The authors declare no conflict of interest. The funders had no role in the design of the study; in the collection, analyses, or interpretation of data; in the writing of the manuscript; or in the decision to publish the results.

References
1. Cornara, D.; Nocera, A.; Corrado, I.; Verrastro, V.; Lamaj, F.; El Kenawy, A.; Russo, V.; Porcelli, F. Lo Zelus renardii (Kolenati, 1857) (Heteroptera Reduviidae): Un promettente predatore della Macrohomotoma gladiata (Kuwayama, 1908) (Psylloidea Homotomidae) sui Ficus microcarpa Hort. Berol. ex Walp. (Moraceae) ornamentali del verde urbano a Bari. In Proceedings of the XXV Congresso Nazionale Entomologia, Padova, Italy, 20–24 June 2016.
2. Cioffi, M.; Cornara, D.; Corrado, I.; Jansen, M.G.M.; Porcelli, F. The status of Aleurocanthus spiniferus from its unwanted introduction in Italy to date. Bull. Insectol. 2013, 66, 273–281.
3. Cornara, D.; Morente, M.; Markheiser, A.; Bodino, N.; Tsai, C.W.; Fereres, A.; Redak, R.A.; Perring, T.M.; Lopes, J.R.S. An overview on the worldwide vectors of Xylella fastidiosa. Entomol. Gen. 2019, 39, 157–181.
4. Picciotti, U.; Lahbib, N.; Sefa, V.; Porcelli, F.; Garganese, F. Aphrophoridae role in Xylella fastidiosa subsp. pauca ST53 invasion in southern Italy. Pathogens 2021, 10, 1035.
5. Acquasanta, F.; Bacci, L.; Baser, N.; Carmignano, P.M.; Cavaliere, V.; Cioffi, M.; Convertini, S.; D’Accolti, A.; Dal Maso, E.; Diana, F.; et al. Tradizione e innovazione nel controllo del Philaenus spumarius Linnaeus, 1758 (Hemiptera Aphrophoridae). In Proceedings of the Giornate Fitopatologiche 2018 Protezione delle Piante, Qualità, Ambiente, Chianciano Terme, Italy, 6–9 March 2018; pp. 181–190.
Insects 2022, 13, 158

Di Serio, F.; Bodino, N.; Cavalieri, V.; Demichelis, S.; Di Carolo, M.; Dongiovanni, C.; Fumarola, G.; Gilioli, G.; Guerrieri, E.; Piccotti, U.; et al. Collection of data and information on biology and control of vectors of Xylella fastidiosa. EFSA Support Publ. 2019, 16, 1628E.

Cormara, D.; Cavalieri, V.; Dongiovanni, C.; Altamura, G.; Palmisano, F.; Bosco, D.; Porcelli, F.; Almeida, R.P.P.; Saponari, M. Transmission of Xylella fastidiosa by naturally infected Philaenus spumarius (Hemiptera, Aphrophoridae) to different host plants. J. Appl. Entomol. 2017, 141, 80–87.

Cuny, A.; Legendre, B.; de Jerphanion, P.; Juteau, V.; Forveille, A.; Germain, J.F.; Ramel, J.M.; Reynaud, P.; Olivier, V.; Poliaikoff, F. Xylella fastidiosa subspecies and sequence types detected in Philaenus spumarius and in infected plants in France share the same locations. Plant Pathol. 2020, 69, 1798–1811.

Scortichini, M. Predisposing Factors for “Olive Quick Decline Syndrome” in Salento (Apulia, Italy). Agronomy 2020, 10, 1445.

Fierro, A.; Liccardo, A.; Porcelli, F. A lattice model to manage the vector and the infection of Xylella fastidiosa on olive trees. Sci. Rep. 2019, 9, 1–14.

Liccardo, A.; Fierro, A.; Garganese, F.; Piccotti, U.; Porcelli, F. A biological control model to manage the vector and the infection of Xylella fastidiosa on olive trees. PLoS ONE 2020, 15, e0232363.

Rosenheim, J.A.; Wilhoit, L.R.; Armer, C.A. Influence of intraguild predation among generalist insect predators on the suppression of an herbivore population. Oecologia 1993, 96, 439–449.

Cisneros, J.J.; Rosenheim, J.A. Ontogenetic change of prey preference in the generalist predator Zelus renardii and its influence on predator–preyterator interactions. Ecol. Monogr. 1997, 22, 399–407.

Cisneros, J.J.; Rosenheim, J.A. Changes in the foraging behavior, within-plant vertical distribution, and microhabitat selection of a generalist insect predator: An age analysis. Environ. Entomol. 1998, 27, 949–957.

Law, Y.H.; Rosenheim, J.A. Effects of combining an intraguild predator with a cannibalistic intermediate predator on a species-level trophic cascade. Ecology 2011, 92, 333–341.

Ambrose, D.P. Biocontrol potential of assassin bugs (Hemiptera: Reduviidae). J. Exp. Zool. 2003, 6, 1–44.

Miller, N.C. The Biology of the Heteroptera; Leonard Hill Ltd.: London, UK, 1956.

Loomans, A.J. Every generalist biological control agent requires a special risk assessment. BioControl 2021, 66, 23–35.

Symondson, W.O.C.; Sunderland, K.D.; Greenstone, M.H. Can generalist predators be effective biocontrol agents? Annu. Rev. Entomol. 2002, 47, 561–594.

Hurd, L.E. Predation: The role of generalist predators in biodiversity and biological control. In Encyclopedia of Entomology; Capinera, J.L., Ed.; Springer: Dordrecht, The Netherlands, 2008; pp. 3038–3042.

Tscharntke, T.; Tylianakis, J.M.; Rand, T.A.; Didham, R.K.; Fahrig, L.; Batáry, P.; Bergtsson, J.; Clough, Y.; Crist, T.O.; Dormann, C.F.; et al. Landscape moderation of biodiversity patterns and processes–eight hypotheses. Biol. Rev. 2012, 87, 661–685.

Piper, M.D. Using artificial diets to understand the nutritional physiology of Drosophila melanogaster. Curr. Opin. Insect Sci. 2017, 23, 104–111.

Grassi, A.; Giorgio, L.; Palmieri, L. Drosophila (Sophophora) suzukii (Matsumura), new pest of soft fruits in Trentino (North-Italy) and in Europe. IOBC WPRS Bull. 2011, 70, 121–128.

Baser, N.; Broutou, O.; Lamaj, F.; Verrastro, V.; Porcelli, F. First finding of Drosophila suzukii (Matsumura) (Diptera: Drosophilidae) in Apulia, Italy, and its population dynamics throughout the year. Fruits 2025, 70, 225–230.

Baser, N.; Broutou, O.; Verrastro, V.; Porcelli, F.; Ioriatti, C.; Anfora, G.; Mazzoni, V.; Rossi Stacconi, M.V. Susceptibility of table grape varieties grown in south-eastern Italy to Drosophila suzukii. J. Appl. Entomol. 2018, 142, 465–472.

Mifsud, D.; Porcelli, F. The psyllid Macrohomotoma gladiata Kuwayama, 1908 (Hemiptera: Psylloidea: Homotomidae): A Ficus pest recently introduced in the EPPO region. EPPO Bull. 2012, 42, 161–164.

Pedata, P.A.; Burchhardt, D.; Mancini, D. Severe infestations of the jumping plant-louse Macrohomotoma gladiata, a new species in Italy for urban Ficus plantations. Bull. Insectol. 2012, 65, 95–98.

Taskin, E. Efficacy of Some Natural Substances on Planococcus fuscus (Signoret) and Their Impact on Its Two Natural Enemies Anagyrus sp. Near pseudococi (Girault) and Cryptolaemus montrouzieri (Mulsant). MSc. Thesis, CIHEAM—Mediterranean Agronomic Institute of Bari, Valenzano, Italy, 2014.

Taskin, E.; Lamaj, F.; Verrastro, V.; Baldacchino, F. Laboratory efficacy of natural substances on Planococcus fuscus (Sign.) and their impact on its two natural enemies. In Proceedings of the Fifth International Scientific Agricultural Symposium Agrosym (Agrosym 2014), Jahorina, Bosnia and Herzegovina, 23–26 October 2014; pp. 483–490.

Hanife, G. Modified agar-based diet for small scale laboratory rearing of olive fruit fly, Bactrocera oleae (Diptera: Tephritidae). Fla. Entomol. 2008, 91, 651–656.

Smout, S.; Asseburg, C.; Matthiopoulos, J.; Fernández, C.; Redpath, S.; Thingood, S.; Harwood, J. The functional response of a generalist predator. PLoS ONE 2010, 5, e10761.

Mahzoum, A.M.; Villa, M.; Benhadi-Marin, J.; Pereira, J.A. Functional response of Chrysoperla carnea (Neuroptera: Chrysopidae) larvae on Saissetia oleae (Olivier)(Hemiptera: Coccidae): Implications for biological control. Agronomy 2020, 10, 1511.

Davranoglou, L.R. Zelus renardii (Kolenati, 1856), a new world redivuid discovered in Europe (Hemiptera: Reduviidae: Harpactorinae). Entomol. Mon. Mag. 2011, 147, 157–162.

Horton, J.R. The citrus thrips. USDA Bull. 1918, 616, 1–42.

Horton, J.R. Argentine ant in relation to citrus groves. USDA Bull. 1918, 647, 1–30.
36. Miranda-Salcedo, M.A.; Loera-Alvarado, E. Fluctuación poblacional de enemigos naturales de trips (Thysanoptera: Thripidae) asociados a limón mexicano (Citrus aurantifolia Swingle) en Michoacán. *Entomol. Mex.* 2019, 6, 151–155.

37. Haviland, D.R.; Beede, R.H.; Daane, K.M. Crop loss relationships and economic injury levels for *Ferrisia gilli* (Hemiptera: Pseudococcidae) infesting pistachio in California. *J. Econ. Entomol.* 2015, 108, 2683–2690.

38. Hicks, V.; Gilder, K.A.; Heinz, K.M. Active predators of crampemyrtle bark scale. In Proceedings of the 63rd Southern Nursery Association Research Conference, Acworth, GA, USA, 12 June 2019; pp.198–201.

39. Garrison, W. New Agricultural Pest for Southern California: Spotted Gum Lerp Psyllid, Eucalyptotalyma Maidenii; San Diego County Agricultural Commissioner’s Office, San Diego, CA, USA; 2001; pp. 1–4.

40. Pereira, J.M. Resistência de Genótipos de Eucalipto ao Psilídeo-de-Concha *Glycaspis brimblecombei* Moore (Hemiptera: Psyllidae). Ph.D. Thesis, Universidade Estadual Paulista “Julio de Mesquita Filho”—UNESP, São Paulo, Brazil, 22 March 2011.

41. Alonso, O. Aspectos fitosanitarios acerca de las plagas insectiles de las arbóreas forrajeras. *Past. Forr.* 2001, 24, 1–18.

42. D’Hervé, F.E.; Olave, A.; Dapoto, G.L. *Zelus renardii* (Hemiptera: Reduviidae: Harpactorinae: Harpactorini): First record from Argentina. *Rev. Soc. Entomol. Arg.* 2018, 27, 32–35.

43. Barrera, J.F.; Gómez-Ruiz, J.; Herrera-Muñoz, J. Biología y Método de Cría de *Zelus renardii* (Hemiptera: Reduviidae), enemigo natural de *Diaphorina citri* (Hemiptera: Psyllidae). In Proceedings of the 1st Simposio Nacional Sobre Investigación Para el Manejo del Psílido Asáctico de los Crípticos y el Huanglongbing en México, Monterrey, Nuevo León, Mexico, 8–9 December 2010; pp. 277–291.

44. Miranda-Salcedo, M.A.; López-Arroyo, J.I.; Velazquez-Monroy, J. Manejo regional de *Diaphorina citri* Kuwayama (Hemiptera: Psyllidae) en Michoacán. In Proceedings of the XLVII Congreso Nacional de Entomología, Benemérita Universidad Autónoma de Puebla Complejo Cultural Universitario, Puebla, México, 21–24 May 2012, 777–782.

45. Albores-Flores, C.I. Detección del ADN de *Diaphorina citri* (Hemiptera: Psyllidae) en el Contenido Intestinal de *Zelus Renardii* (Hemiptera: Reduviidae). MSc. Thesis, El Colegio de la Frontera Sur, Lerma, Campeche, Mexico, 2016.

46. Miranda-Salcedo, M.A.; Pardo-Melgar, S. Manejo del psílido asiático de los citricos *Diaphorina citri* Kuwayama (Hemiptera: Psyllidae) en Michoacán. In Proceedings of the XXXVIII Congreso Nacional de Control Biológico, Sociedad Mexicana de Control Biológico, León de los Aldama, Guanajuato, Mexico, 5–6 November 2015; pp. 203–208.

47. Miranda-Salcedo, M.A. Manejo en areas regionales del control de *Diaphorina citri* Kuwayama, 1908 (Hemiptera: Psyllidae) en Michoacan. *Entomol. Mex.* 2018, 5, 342–347.

48. Hagler, J.R.; Naranjo, S.E. Determining the frequency of heteropteran predation on sweetpotato whitefly and pink bollworm using multiple ELISAs. *Entomol. Exp. Appl.* 1994, 72, 59–66.

49. Naranjo, S.E.; Ellsworth, P.C.; Chu, C.C.; Henneberry, T.J. Conservation of predatory arthropods in cotton: Role of action thresholds for *Bemisia tabaci* (Homoptera: Aleyrodidae). *J. Econ. Entomol.* 2002, 95, 682–691.

50. Naranjo, S.E.; Ellsworth, P.C.; Hagler, J.R. Conservation of natural enemies in cotton: Role of insect growth regulators in management of *Bemisia tabaci*. * Biol. Control* 2004, 30, 52–72.

51. Hagler, J.R.; Blackmer, F. Identifying inter- and intra-guild feeding activity of an arthropod predator assemblage. *Ecol. Entomol.* 2013, 38, 258–271.

52. Nielson, M.W.; Henderson, J.A. Biology of *Collops vittatus* (Say) in Arizona, and feeding habits of seven predators of the spotted alfalfa aphid. *J. Econ. Entomol.* 1959, 52, 159–162.

53. Fye, R.E. Cotton insect populations: Development and impact of predators and other mortality factors [Arizona]. USDA Technical Bull. 1979, 1592, 1–65.

54. Rosenheim, J.A.; Kaya, H.K.; Ehler, L.E.; Marois, J.J.; Jaffée, B.A. Intraguild predation among biological-control agents: Theory and evidence. *Biol. Control* 1995, 5, 303–335.

55. Rosenheim, J.A.; Limburg, D.D.; Colfer, R.G. Impact of generalist predators on a biological control agent, Chrysoperla carnea: Direct observations. *Ecol. Appl.* 1999, 9, 409–417.

56. Nelson, E.H.; Rosenheim, J.A. Encounters between aphids and their predators: The relative frequencies of disturbance and consumption. *Entomol. Exp. Appl.* 2006, 118, 211–219.

57. Potin Téllez, C.A. Tabla de Vida Del Depredador *Zelus Renardii* (Kolenati) (Hemiptera: Heteróptera: Reduviidae) en Laboratorio. MSc. Thesis, Universidad de Chile, Santiago, Chile, 2008.

58. El-Tom, H.A. The biology of *Zelus renardii* Kolenati and *Zelus socius* Uhler (Hemiptera: Reduviidae). Ph.D. Thesis, University of Arizona, Tucson, AZ, USA, 15 January 1965.

59. Hagler, J.; Groves, R.; Johnson, M.W.; Morgan, D. An immunological approach for quantifying predation rates on the glassy-winged sharpshooter. In Proceedings of the Pierce’s Disease Research Symposium, San Diego, CA, USA, 27–29 November 2006; pp. 70–72.

60. Fournier, V.; Hagler, J.R.; Daane, K.; De León, J.; Groves, R. Identifying the predator complex of *Homalodisca vitripennis* (Hemiptera: Cicadellidae): A comparative study of the efficacy of an ELISA and PCR gut content assay. *Oecologia* 2008, 157, 629–640.

61. Hagler, J.; Groves, R.; Johnson, M.W.; Morgan, D. Pinpointing predation events: A different molecular approach. In Proceedings of the Third International Symposium on Biological Control of Arthropods (ISBCA 3) Christchurch, New Zealand, 8–13 February 2009; pp. 461–467.
62. Salerno, M.; Russo, V.; Sefa, V.; Lamaj, F.; Bashier, N.; Verrastro, V.; Porcelli, F. *Zelus renardii* an assassin bug candidate for *Philaenus spumarius* biocontrol. In Proceedings in the European Conference on *Xylella* Finding Answer to a Global Problem, Palma de Mallorca, Spain, 13–15 November 2017; pp. 22–23.

63. Kron, C.R.; Sisterson, M.S. *Spissistilus festinus* (Hemiptera: Membracidae) susceptibility to six generalist predators. *PLoS ONE 2020*, 15, e0242775.

64. Muir, F. The sugar cane leafhopper and its parasites in Hawaii. *Hawaiian Planters’ Record 1921*, 25, 108–123.

65. Nishida, T. Natural enemies of the melon fly, *Dacus cucurbitae* Coq. in Hawaii. *Ann. Entomol. Soc. Am. 1955*, 48, 171–178.

66. Risco, S.H. Notas adicionales sobre el “saltahoja” de la caña de azúcar *Perkinsiella saccharicida* K. *Rev. Peru. Entomol. 1969*, 9, 181–187.

67. Ali, A.S.A. Predator-Prey Relationship of *Zelus renardii* (Kolenati) and *Bucculatrix thurberiella* (Busck). Master’s Thesis, University of Arizona, Tucson, AZ, USA, 1978; p. 98.

68. Weirauch, C. Observations on the sticky trap predator *Zelus laridus* Stål (Heteroptera, Reduviidae, Harpactorinae), with the description of a novel gland associated with the female genitalia. *Denisia 2006*, 19, 1169–1180.

69. Fullaway, D.T. The corn leaf hopper (*Peregrinus maidis*, Ashm.). *Bull. Hawaii Entomol. 1918*, 4, 1–16.

70. Swezey, O.H. Biological control of the sugar cane leafhopper in Hawaii. *Hawaiian Sugar Planters’ Assoc. Exp. Sta. Bull. Entomol. Ser. 1936*, 21, 57–101.

71. Zimmerman, E.C. Insects of Hawaii, vol. 3. Heteroptera. Bernice, P. Bishop Museum; Experiment Station, Hawaiian Sugar Planters’ Association: Honolulu, HI, USA, 1948.

72. Napompeth, B. Ecology and Population Dynamics of the Corn Plantthopper, *Peregrinus maidis* (Ashmead) (Hemiptera: Delphladdockae), in Hawaii. Ph.D. Thesis, University of Hawaii, Honolulu, HI, USA, 1973.

73. Holdaway, F.G.; Loc, W.C. Insects of the garden bean in Hawaii. *Proc. Hawaii Entomol. Soc. 1942*, 11, 249–260.

74. Hagler, J.R.; Allen, C.C.; Bradley-Dunlop, D.; Enriquez, F.J. Field evaluation of predation on *Lygus hesperus* (Hemiptera: Miridae) using a species-and stage-specific monoclonal antibody. *Environ. Entomol. 1992*, 21, 896–900.

75. Hagler, J.R. Development of an immunological technique for identifying multiple predator–prey interactions in a complex arthropod assemblage. *Ann. Appl. Biol. 2006*, 149, 153–165.

76. Zink, A.G.; Rosenheim, J.A. Stage-specific predation on *Lygus hesperus* affects its population stage structure. *Entomol. Exp. Appl. 2008*, 126, 61–66.

77. Assimwe, P.; Naranjo, S.E.; Ellsworth, P.C. Effects of irrigation levels on interactions among *Lygus hesperus* (Hemiptera: Miridae), insecticides, and predators in cotton. *Environ. Entomol. 2014*, 43, 263–273.

78. Rosenheim, J.A. Source–sink dynamics for a generalist insect predator in habitats with strong higher-order predation. *Ecol. Monogr. 2001*, 71, 93–116.

79. Greemstone, M.H.; Tillman, P.G.; Hu, J.S. Predation of the newly invasive pest *Megacopta cribraria* (Hemiptera: Plataspidae) in soybean habitats adjacent to cotton by a complex of predators. *J. Econ. Entomol. 2014*, 107, 947–954.

80. Lahiri, S.; Reigis, D.D. Ecology and management of kudzu bug (Hemiptera: Plataspidae) in Southeastern soybeans. *J. Integr. Pest Manag. 2016*, 7, 1–6.

81. Grant, J.I. Phenology and Cold Tolerance of *Megacopta cribraria*: An Invasive Soybean Pest at Its Northern Limit. M.Sc. Thesis, University of Maryland, College Park, MA, USA, 2016.

82. Morrill, A.W. Plant-bugs injurious to cotton bolls. *USDA 1910*, 86, 1–110.

83. Tillman, P.G.; Greemstone, M.H.; Hu, J.S. Predation of stink bugs (Hemiptera: Pentatomidae) by a complex of predators in cotton and adjoining soybean habitats in Georgia, USA. *Fla. Entomol. 2015*, 98, 1114–1126.}

84. Brown, L.M. Biology, Ecology, and Economics of Brown Stink Bug, *Euschistus servus* (Heteroptera: Pentatomidae), in Desert Cotton Agroecosystems. M.Sc. Thesis, University of Arizona, Tucson, AZ, USA, 2017; p. 75.

85. Clancy, D.W. Natural enemies of some Arizona cotton insect. *J. Econ. Entomol. 1946*, 39, 326–328.

86. Ehler, L.E. An evaluation of some natural enemies of *Nezara viridula* in northern California. *BioControl 2002*, 47, 309–325.

87. Knowlton, G.F. Observation on the feeding of some predaceous Hemiptera. *Proc. Utah Acad. Sci. 1944*, 21, 37–39.

88. Rosenheim, J.; Willhoit, L. Why lacewings may fail to suppress aphids... Predators that eat other predators disrupt cotton aphid control. *Calif. Agric. 1993*, 47, 7–9.

89. Ehler, L.E.; Long, R.F.; Kinsey, M.G.; Kelley, S.K. Potential for augmentative biological control of black bean aphid in California sugarbeet. *Entomophaga 1997*, 42, 241.

90. Ambrose, D.P. Assassin bugs (Reduviidae excluding Triatominae). In *Heteroptera of Economic Importance*; Schaefer, C.W., Panizzi, A.R., Eds.; CRC Press LLC: Boca Raton, FL, USA, 2000; pp. 695–712.

91. Hagler, J.R.; Blackmer, F.; Spurgeon, D.W. Accuracy of a prey-specific DNA assay and a generic prey-immunomarking assay for detecting predation. *Methods Ecol. Evol. 2015*, 6, 1426–1434.

92. Hagler, J.R.; Blackmer, F. Evidence of intraguild predation on a key member of the cotton predator complex. *Food Webs 2015*, 4, 8–13.

93. Yaherwandi, Y.; Diratika, M. Kelimpahan kepik predator (Hemiptera: Reduviidae) di daerah endemik serangan ulat api pada perkebunan kelapa sawit rakyat. *J. Penelitian Pertanian Terapan 2020*, 20, 1–10.

94. Tuttle, D.M.; Wene, G.P.; Sheets, L.W. The cotton leaf perforator and its control in Arizona. *J. Econ. Entomol. 1961*, 54, 67–70.

95. Orphanides, G.M.; Gonzalez, D.; Bartlett, B.R. Identification and evaluation of pink bollworm predators in southern California. *J. Econ. Entomol. 1971*, 64, 421–424.
96. Sarwar, M. Biological parameters of pink bollworm *Pectinophora gossypiella* (Saunders) (Lepidoptera: Gelechiidae): A looming threat for cotton and its eradication opportunity. *Int. J. Res. Agric. Forest.* 2017, 4, 25–36.

97. Atkins Jr., E.L.; Frost Jr., M.H.; Anderson, L.D.; Deal, A.S. The “omnivorous leaf roller,” *Platynota stultana* Wshlm., on cotton in California: Nomenclature, life history, and bionomics (Lepidoptera, Tortricidae). *Ann. Entomol. Soc. Am.* 1957, 50, 251–259.

98. Horton, J.R. A swallowtail butterfly injurious to California orange trees. *Mon. Bull. Calif. State Hortic. Comm.* 1922, 11, 377–387.

99. Ewing, K.P.; Ivy, E.E. Some factors influencing bollworm populations and damage. *J. Econ. Entomol.* 1943, 36, 602–606.

100. Lingren, P.D.; Ridgway, R.L.; Jones, S.L. Consumption by several common arthropod predators of eggs and larvae of two Heliotis species that attack cotton. *Ann. Entomol. Soc. Am.* 1968, 61, 613–618.

101. Ali, A.S.A.; Watson, T.F. Effect of temperature on development and survival of *Zelus renardii*. *Environ. Entomol.* 1978, 7, 889–890.

102. Cohen, A.C. Organization and preliminary characterization of salivary trypsin-like enzymes in a predaceous heteropteran, *Zelus renardii*. *J. Insect Physiol.* 1993, 39, 823–829.

103. Shrestha, R.B.; Parajulee, M.N.; Blanco, C. Functional response of selected cotton arthropod predators to bollworm eggs in the laboratory. In Proceeding of Beltwide Cotton Conference, San Antonio, TX, USA, 5–9 January 2004; pp. 5–9.

104. Wille, J.E. Biological control of certain cotton insects and the application of new organic insecticides in Peru. *J. Econ. Entomol.* 1951, 44, 13–18.

105. Ables, J.R. Feeding behavior of an assassin bug, *Zelus renardii*. *Ann. Entomol. Soc. Am.* 1978, 71, 476–478.

106. Cohen, A.C.; Tang, R. Relative prey weight influences handling time and biomass extraction in *Sinea confusa* and *Zelus renardii* (Heteroptera: Reduviidae). *Environ. Entomol.* 1997, 26, 559–565.

107. Ehler, L.E. An evaluation of some natural enemies of *Spodoptera exigua* on sugarbeet in northern California. *BioControl* 2004, 49, 121–135.

108. Eisenring, M.; Romeis, J.; Naranjo, S.E.; Meissle, M. Multitrophic Cry-protein flow in a dual-gene Bt-cotton field. *Agric. Ecosyst. Environ.* 2017, 247, 283–289.

109. Bisabri-Ershadi, B.; Ehler, L.E. Natural biological control of western yellow-striped armyworm, *Spodoptera praefera* (Grote) in hay alfalfa in northern California. *Hilgardia* 1981, 49, 1–23.

110. Su, H.H.; Tian, J.C.; Naranjo, S.E.; Romeis, J.; Hellmich, R.L.; Shelton, A.M. Bacillus thuringiensis plants expressing Cry1Ac, Cry2Ab and Cry1F are not toxic to the assassin bug, *Zelus renardii*. *J. Appl. Entomol.* 2015, 139, 23–30.

111. Van Zwaluwenburg, R.H. Recent immigrant insects. *Hawaii. Plant. Rec.* 1946, 50, 11–17.

112. Mbata, K.J.; Hart, E.R.; Lewis, R.K. Reproductive behavior in *Zelus renardii* Kolenati 1857 (Hemiptera: Reduviidae). *Iowa State J. Res.* 1987, 62, 261–265.

113. Severin, H.H.; Severin, H.C.; Hartung, W.J. The ravages, life history, weights of stages, natural enemies and methods of control of the melon fly (Dacus cucurbitae Coq.). *Ann. Entomol. Soc. Am.* 1914, 7, 177–207.

114. Faúndez, E.I. La chinche asesina *Zelus renardii* (Kolenati, 1856) (Heteroptera: Reduviidae) en Chile: Comentarios después de 15 años de su llegada al país. *Bol. Soc. Entomol. Arag.* 2015, 57, 421–423.

115. Hinckley, A.D. The klu beetle, *Mimoseses sallaei* (Sharp), in Hawaii (Coleoptera: Bruchidae). *Proc. Hawaii Entomol. Soc.* 1960, 17, 260–269.

116. Campbell, R.E. The broad-bean weevil. *USDA Bull.* 1920, 807, 1–23.

117. Mezani, S. Bioécologie de la Bruche de la Fève *Bruchus rufimanus* Boh. (Coleoptera: Bruchidae) dans des Parcelles de variétés de Fèves différentes et de féverole dans la région de Tizi-Rached (Tizi-Ouzou). Master’s Thesis, University Mouloud Mammeri de Tizi Ouzou, Tizi Ouzou, Algeria, 2011.

118. Mezani, S. Suivi des populations de *Bruchus rufimanus* (Coleoptera: Chrysomelidae) dans les lieux de diapause et dans des parcelles de variétés de Fèves différentes (Aguadulce; Séville et Féverole) dans la région de Tizi-Ouzou. Ph.D. Thesis, University Mouloud Mammeri de Tizi Ouzou, Tizi Ouzou, Algeria, 2016.

119. Moran, P.J. Lack of establishment of the Mediterranean tamarisk beetle *Diorhabda elongata* (Coleoptera: Chrysomelidae) on athel (*Tamarix aphylla*) (Tamaricaceae) in South Texas. *Southwest Entomol.* 2010, 35, 129–145.

120. Hernandez, M.; Masonick, P.; Weirauch, C. Crowdsourced online images provide insights into predator-prey interactions of putative natural enemies. *Food Webs* 2019, e00126.

121. Drees, B.M.; Jackman, J. Field guide to Texas Insects. Texas A&M Agrilife Extension (blog). 1999. Available online: https://texasinsects.tamu.edu/ (accessed 31 August 2021).

122. Muir, F.A.G. Introduction. In *Handbook of the Insects and Other Invertebrates of Hawaiian Sugarcane Fields*; Williams, F.X., Ed.; Experiment Station of the Hawaiian Sugar Planters’ Association: Honolulu, HI, USA, 1931; pp. 11–32.

123. Curkovik, T.; Araya, J.E.; Baena, M.; Guerrero, M.A. Presencia de *Zelus renardii* Kolenati (Heteroptera: Reduviidae) en Chile. *Bol. Soc. Entomol. Arag.* 2004, 34, 163–165.

124. Heimpe1, G.E.; Rosenheim, J.A.; Mangel, M. Predation on adult *Aphytis* parasitoids in the field. *Oecologia* 1997, 110, 346–352.

125. Cogni, R.; Freitas, A.V.L.; Amaral Filho, B.F. Influence of prey size on predation success by *Zelus longipes* L. (Heti., Reduviidae). *J. Appl. Entomol.* 2002, 126, 74–78.

126. Zhang, G.; Hart, E.R.; Weirauch, C. A taxonomic monograph of the assassin bug genus *Zelus* Fabricius (Hemiptera: Reduviidae): 71 species based on 10,000 specimens. *Biodivers. Data J.* 2016, 4, e8150.

127. van der Heyden, T. First records of *Zelus renardii* (Kolenati, 1856) (Hemiptera: Heteroptera: Reduviidae: Harpactorinae) for Albania. *Arq. Entomol.* 2017, 49–50.
Insects 2022, 13, 158

128. Sahayaraj, K. Reduviids and Their Merits in Biological Control. In Basic and Applied Aspects of Biopesticides; Sahayaraj, K., Ed.; Springer: New Delhi, India, 2014; pp. 195–214.

129. Sahayaraj, K.; Balasubramanian, R. Reduvid: An important biological control agent. In Artificial Rearing of Reduvid Predators for Pest Management; Sahayaraj, K.; Balasubramanian, R., Eds; Springer: New Delhi, India, 2016; pp. 1–28.

130. Weirauch, C.; Alvarez, C.; Zhang, G. Zelus renardii and Z. tetracanthus (Hemiptera: Reduviidae): Biological attributes and the potential for dispersal in two assassin bug species. Fla. Entomol. 2012, 95, 641–649.

131. Kolenati, F.A. Meletemata Entomologica Hemipterorum Heteropterorum Caucasi. Harpagocorisiae, Monographice Dispositae. Fasciculum IV. Bull. Soc. Nat. Moscou 1856, 1–502.

132. Henry, T.J. Biodiversity of Heteroptera. In Insect Biodiversity: Science and Society; Foottit, R.G., Adler, P.H., Eds.; Wiley-Blackwell: Oxford, UK, 2009; pp. 223–254.

133. Baena, M.; Torres, J.L. Nuevos datos sobre heterópteros exóticos en España y Francia: Temmyra biguttula Stål, 1874, Belonochilus numenius (Say, 1832) y Zelus renardii (Kolenati, 1856) (Heteroptera: Rhypharachnidae, Orsillidae, Reduviidae). Bol. Assoc. Esp. Entomol. 2012, 36, 351–360.

134. Vivas, L. Primera cita en España de la especie Zelus renardii (Kolenati, 1857) (Heteroptera: Reduviidae) que representa la segunda cita en Europa. BVnPC 2012, 1, 34–40.

135. Dioli, P. Zelus renardii (Kolenati, 1856) (Insecta Heteroptera Reduviidae). Quad. Studi Nat. Romagna 2013, 38, 232–233.

136. Puchtkov, P.V. Invasive true bugs (Heteroptera) established in Europe. Ukr. Entomol. J. 2013, 2, 11–28.

137. Pinzari, M.; Cianferoni, F.; Martello, S.; Dioli, P. Zelus renardii (Kolenati, 1856), a newly established alien species in Italy (Hemiptera: Reduviidae, Harpactorinae). Fragn. Entomol. 2018, 50, 31–35.

138. Swezey, O.H. Leaf-hoppers and their natural enemies (Pt. VII. Orthoptera, Coleoptera, Hemiptera). Rept. Expt. Sta. Hawaiian Sugar Planters Assoc. Bull. Div. Entomol. 1905, 7, 209–238.

139. Kirkaldy, G.W. Biological notes on the Hemiptera of the Hawaiian Isles No. 1. Proc. Hawaii Entomol. Soc. 1907, I, 135–161.

140. Kirkaldy, G.W. On some Hawaiian Hemiptera-Hemiptera. Can. Entomol. 1907, 39, 244–248.

141. Perkins, R.C.L. Introduction being a review of the land-fauna of Hawaii. In Fauna Hawaïennise or the Zoology of the Sandwich (Hawaiian) Isles; Sharp, D., Ed.; Cambridge University Press: Cambridge, UK, 1913; pp. 1–16.

142. Hart, E.R. A Systematic Revision of the Genus Zelus Fabricius (Hemiptera: Reduviidae). Ph.D. Thesis, Texas A&M University, College Station, TX, USA, 1972.

143. Hart, E.R. Genus Zelus Fabricius in the United States, Canada, and northern Mexico (Hemiptera: Reduviidae). Ann. Entomol. Soc. Am. 1986, 79, 535–548.

144. Hart, E.R. The genus Zelus Fabricius in the West Indies (Hemiptera: Reduviidae). Ann. Entomol. Soc. Am. 1987, 80, 293–305.

145. Werner, F.G.; Butler, J.R. The rediviids and nabids associated with Arizona crops. Univ. Ariz. Agr. Exp. Sta. Tech. Bul. 1957, 133, 1–12.

146. Çerçi, B.; Koçak, Ö. Contribution to the knowledge of Heteroptera (Hemiptera) fauna of Turkey. J. Insect Biodivers. 2016, 4, 1–18.

147. Van der Heyden, T. First record of Zelus renardii Kolenati (Heteroptera: Reduviidae: Harpactorinae) in Israel. Rev. Chil. Entomol. 2018, 44, 463–465.

148. Kök, Ş.; Tomanovic, Ž.; Nedeljkovic, Z.; Šenal, D.; Kasap, İ. Biodiversity of the natural enemies of aphids (Hemiptera: Aphididae) in Northwest Turkey. Phytoparasitica 2020, 48, 51–61.

149. Kiyak, S. The new record invasive alien species (IAS) Zelus renardii (Kolenati, 1856) (Hemiptera: Heteroptera: Reduviidae) in central Anatolia (Turkey). J. Hid. Turk. 2020, 2, 47–52.

150. Perkins, R.C.L. The introduction of beneficial insects into the Hawaiian Islands. Nature 1897, 55, 499–500.

151. Pettrakis, P.V.; Moulet, P. First record of the nearctic Zelus renardii (Heteroptera, Reduviidae, Harpactocorinae) in Europe. Entomol. Hell. 2011, 20, 75–81.

152. Simov, N.; Gradinarov, D.; Davranoglou, L.R. Three new assassin bug records (Hemiptera: Heteroptera: Reduviidae) for the Balkan Peninsula. Ecol. Montenegro 2017, 13, 25–29.

153. Van der Heyden, T. A recent record of Zelus renardii (Kolenati, 1856) on Crete/Greece (Hemiptera: Heteroptera: Reduviidae: Harpactorinae). BVnPC 2015, 4, 55–59.

154. Battaglia, D.; Mele, G. La cimice assassina, Zelus renardii, in Basilicata. Agrifoglio 2020, 98, 1–5.

155. Rattu, A.; Dioli, P. Prima segnalazione di Zelus renardii (Kolenati, 1856) in Sardegna (Hemiptera, Reduviidae). Ricerca Gal. Entomol. 2020, 11, 119–125.

156. Bella, S. The Nearctic bug Zelus renardii (Kolenati) (Hemiptera Reduviidae) in northern Italy and Sicily. J. Zool. 2020, 103, 87–88.

157. Garrouste, R. Zelus renardii (Kolenati, 1856): Une réduve nouvelle pour la France (Hemiptera, Reduviidae, Harpactorinae). Bull. Soc. Entomol. Fr. 2019, 124, 335–336.

158. Van der Heyden, T.A.; Grosso-Silva, J.M. First record of Zelus renardii Kolenati, 1856 in Portugal (Heteroptera: Reduviidae: Harpactorinae). Arq. Entomol. 2020, 22, 347–349.

159. Van der Heyden, T.A. Erstfund von Zelus renardii Kolenati, 1856 in Deutschland (Heteroptera: Reduviidae). Heteropteron 2021, 61, 31–32.

160. Van der Heyden, T.A. On the recent northern European dispersion of Zelus renardii Kolenati (Hemiptera: Heteroptera: Reduviidae) via human activity. Isr. J. Entomol. 2021, 51, 43–46.
161. Rodríguez-Lozano, B.; Gómez-de Dios, M.Á. The invasive species Zelus renardii (Kolenati, 1857) (Hemiptera, Reduviidae) in Spain and comments about its global expansion. *Trans. Am. Entomol. Soc.* 2018, 144, 551–558.

162. Goula, M.G.; Lizana, F.; Núñez, A.M. New records of the Nearctic leafhopper assassin bug, Zelus renardii Kolenati, 1857 in the Iberian Peninsula (Hemiptera: Heteroptera: Reduviidae). *Bull. Inst. Catalana Hist. Nat.* 2019, 83, 219–222.

163. Pérez-Gómez, Á.; Sánchez, I.; Baena, M. Nuevos registros de hemípteros (Insecta: Hemiptera) alóctonos en Andalucía (sur de España). *Rev. Soc. Lad. Hist. Nat.* 2020, 14, 9–19.

164. Pyšek, P.; Richardson, D.M. Traits associated with invasiveness in alien plants: Where do we stand? In *Biological Invasions*; Nentwig, W., Ed.; Springer: Berlin/Heidelberg, Germany, 2007; pp. 97–125.

165. Van Kleunen, M.; Dawson, W.; Schlaepfer, D.; Jeschke, J.M.; Fischer, M. Are invaders different? A conceptual framework of comparative approaches for assessing determinants of invasiveness. *Ecol. Lett.* 2010, 13, 947–958.

166. Whitcomb, W.H.; Bell, K.O. Predaceous insects, spiders, and mites of Arkansas cotton fields. *Agric. Exp. Sta. Div. of Agric. Univ. Ark.* 1964, 690, 1–84.

167. Hagler, J.R; Casey, M.T.; Machtley, S.A. A procedure for pinpointing cannibalism, intraguild predation, and life stage-specific feeding events. *BioControl* 2020, 1–8.

168. Stoner, A.; Metcalfe, A.M.; Weeks, R.E. Plant feeding by Reduviidae, a predaceous family (Hemiptera). *J. Kans. Entomol. Soc.* 1975, 48, 185–188.

169. Van den Bosch, R.; Hagen, K.S. Predaceous and parasitic arthropods in California cotton fields. *Calif. Agric. Exp. Sta. Bull.* 1966, 820, 1–32.

170. Law, Y.H.; Sediqi, A. Sticky substance on eggs improves predation success and substrate adhesion in newly hatched Zelus renardii (Hemiptera: Reduviidae) instars. *Ann. Entomol. Soc. Am.* 2010, 103, 771–774.

171. Dejean, A.; Revel, M.; Azémar, F.; Roux, O. Altruism during predation in an assassin bug. *Naturwissenschaften* 2013, 100, 913–922.

172. Smith, G.L. *California Cotton Insects*; University of California Experiment Station: Berkeley, CA, USA, 1942.

173. Ambrose, D.P.; Ambrose, A.D. Predation, copulation, oviposition and functional morphology of tibia, rostrum and eggs as tools in the biosystematics of Reduviidae (Hemiptera). *Indian J. Entomol.* 2009, 71, 1–17.

174. Zhang, G.; Weirauch, C. Sticky predators: A comparative study of sticky glands in harpactorine assassin bugs (Insecta: Hemiptera: Reduviidae). *Acta Zool.* 2013, 94, 1–10.

175. Cohen, A.C. Feeding adaptations of some predaceous Hemiptera. *Ann. Entomol. Soc. Am.* 1990, 83, 1215–1223.

176. Greenop, A. Using Species Traits to Understand the Mechanisms Driving Pollination and Pest Control Ecosystem Services. Ph.D. Thesis, Lancaster University, Lancaster, UK, 2020.

177. Force, D.C. r-and K-strategists in endemic host-parasitoid communities. *Bull. Entomol. Soc. Am.* 1972, 18, 135–137.

178. Greenstone, M.H. Spider feeding behaviour optimises dietary essential amino acid composition. *Nature* 1979, 282, 501–503.

179. Carey, J.R. Insect biodemography. *Annu. Rev. Entomol.* 2001, 46, 79–110.

180. Peterson, R.K.; Davis, R.S.; Higley, L.G.; Fernandes, O.A. Mortality risk in insects. *Nature* 1979, 281, 913–922.

181. Plagans, M.J.; Whitcomb, W.H. Corrn residue as an overwintering site for spiders and predaceous insects in Florida. *Fla. Entomol.* 1986, 69, 665–671.

182. Foster, S.P.; Harris, M.O. Behavioral manipulation methods for insect pest-management. *Annu. Rev. Entomol.* 1997, 42, 123–146.

183. Cook, S.M.; Khan, Z.R.; Pickett, J.A. The use of push-pull strategies in integrated pest management. *Annu. Rev. Entomol.* 2007, 52, 375–400.

184. Mafra-Neto, A.; de Lame, F.M.; Munson, A.S.; Perring, T.M.; Stelinski, L.L.; Mafra, L.E.J.; Borges, R.; Vargas, R.I. Manipulation of insect behavior with specialized pheromone and lure application technology (SPLAT®). In *Pest Management with Natural Products*; Beck, J.J., Coats, J.R., Duke, S.O., Koivunen, M., Ed.s; American Chemical Society: Columbus, OH, USA, 2013; pp. 31–58.

185. Fand, B.B.; Amala, U.; Yadav, D.S.; Rathi, G.; Mhaske, S.; Upadhyay, A.; Ahammed Shabeer, T.P.; Kumbhar, D.R. Bacterial volatiles from mealybug honeydew exhibit kairomonal activity toward solitary endoparasitoid *Anagrus dactylopii*. *J. Pest Sci.* 2020, 93, 195–206.

186. Lozano-Soria, A.; Picciotti, U.; Lopez-Moya, F.; Lopez-Cepero, J.; Porcelli, F.; Lopez-Llorca, L.V. Volatile organic compounds from entomopathogenic and nematophagous fungi, repel banana black weevil (*Cosmopolites sordidus*). *Insects* 2020, 11, 509.

187. Orzali, L.; Mosconi, F.; Roversi, P.F.; Bergamaschi, V.; Riccioni, L. First report of the invasive pest *Drosophila suzukii* (Matsumura, 1931) (Diptera: Drosophilidae) on *Olea europaea* L. in Italy. *REDIA* 2020, 103, 109–112.

188. Nugnes, F.; Laudon, S.; Jesu, G.; Jansen, M.G.M.; Bernardu, U.; Porcelli, F. *Aleocharus spiniferus* (Hemiptera: Aleyrodidae) in some European countries: Diffusion, hosts, molecular characterization, and natural enemies. *Insects* 2020, 11, 42.

189. Bubici, G.; Prigigallo, M.I.; Garganese, F.; Nugnes, F.; Jansen, M.G.M.; Porcelli, F. First report of *Aleocharus spiniferus* on *Ailanthus altissima*: Profiling of the insect microbiome and microRNAs. *Insects* 2020, 11, 161.

190. Wolf, K.W.; Reid, W. Surface morphology of legs in the assassin bug *Zelus longipes* (Hemiptera: Reduviidae): A scanning electron microscopy study with an emphasis on hairs and pores. *Ann. Entomol. Soc. Am.* 2001, 94, 457–461.

191. Fischer, P.; Grozinger, C.M. Phenomenal regulation of starvation resistance in honey bee workers (*Apis mellifera*). *Naturwissenschaften* 2008, 95, 723–729.

192. Strangways-Dixon, J. Hormonal control of selective feeding in female *Calliphora erythrocephala* Meig. *Nature* 1959, 184, 2040–2041.
193. House, H.L. Insect nutrition. *Ann. Rev. Biochem.* **1962**, *31*, 653–672.

194. Robinson, W.H. Urban insects and arachnids. In *A Handbook of Urban Entomology*; Cambridge University Press: Cambridge, UK, 2005.

195. Miralles-Núñez, A.; Pradera, C.; Pujol-Fructuoso, J.A. La problemática de las especies exóticas: El caso de las picaduras producidas por *Zelus renardii* Kolenati, 1857 (Hemiptera: Reduviidae) en España. *Arg. Entomol.* **2021**, *24*, 133–138.

196. Pereira-dos Santos, C.E.; de Souza, J.R.; Zanette, R.A.; da Silva, F.J.; Strussmann, C. Bite caused by the assassin bug *Zelus Fabricius*, 1803 (Hemiptera; Heteroptera: Reduviidae) in a human. *Wilderness Environ. Med.* **2019**, *30*, 63–65.

197. Rassi Jr., A.; Rassi, A.; Marin-Neto, J.A. Chagas disease. *Lancet* **2010**, *375*, 1388–1402.

198. Jurberg, J.; Galvão, C.; Weirauch, C.; Moreira, F.F.F. Hematophagous bugs (Reduviidae, Triatominae). In *True bugs (Heteroptera) of the Neotropics. Entomology in Focus*; Panizzi, A., Grazia, J., Eds.; Springer: Dordrecht, The Netherlands, 2015; Volume 2, pp. 353–393.