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Arthropod pests and their management, natural enemies and flora visitors associated with castor (*Ricinus communis*), a biofuel plant: a review

Artrópodos plaga y su manejo, enemigos naturales y visitantes florales asociados a la higuerilla (*Ricinus communis*), un cultivo bioenergético: revisión

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Abstract: Interest in bioenergetic crops, such as the castor oil plant *Ricinus communis* (Euphorbiaceae), for production of biodiesel has increased in recent years. In this paper, phytophagous arthropods, their natural enemies and floral visitors associated with this plant in the world are reviewed. Despite its insecticidal properties, arthropods have been reported feeding on *R. communis* plants. The arthropod pests of *R. communis* damage all parts of the plant, including the seeds, where some toxic compounds are even more concentrated. In the scientific databases, we found reports of 193 arthropods associated to *R. communis* in different parts of the world. This information obtained in the scientific databases was concentrated in a database and analyzed according to the coevolutive hypothesis, which allows us to predict that the greatest wealth and abundance of phytogenic arthropods is found in the center of origin by *R. communis*. According to this review, *Achaea janata*, *Spodoptera litura*, *Edwardsiana flavescens*, *Liriomyza trifolii*, *L. sativae*, *Spilosoma obliqua*, *Cogenethes punctiferalis*, *Oxyrhachis taranda*, and *Helicoverpa armigera* are the most devastating pests in Asia. In Africa, *Agrotis ipsilon*, *S. exigua*, *Nezara viridula*, *Trialeurodes ricini*, and *Tetranychus urticae* were mentioned as the most important. In Central and South-America, *Phyllophaga* sp., *Agrietes* sp., *Erinnyis ello*, *N. viridula, Corythucha gossypii*, *Falconia antioquiana*, and *S. marima* are reported as pests of economic importance. The most commonly reported natural enemies of some of these arthropod pests were species of *Bacillus thuringiensis, B. cereus, B. popilliae, Trichogramma achaeae, T. chilonis, T. minutum, T. australicum, T. dendrolimi, T. pretiosum, T. evanescens, Microplitis rufiventris, M. maculipennis, M. ophiusae, Telenomus remus, T. proditor, Stethorus siphonulus* and *S. histrio*. *Apis mellifera* is recorded as the main insect pollinator of *R. communis*. Pest management methods used against the arthropod pests of *R. communis* include biological, ethological, mechanical, cultural, genetic, and chemical control.

Keywords: Castor-oil plant, biodiesel, pests, entomophagous organisms, pollinators.

Resumen: El interés por los cultivos bioenérgeticos, tales como *Ricinus communis* (Euphorbiaceae) para producir biodiesel ha aumentado en años recientes. En este documento se hace una revisión sobre los artrópodos fitófagos, enemigos naturales y visitantes florales asociados a esta planta en el mundo. A pesar de las propiedades insecticidas de *R. communis*, existen registros sobre artrópodos que se alimentan de ella. Los artrópodos plaga de *R. communis* dañan toda la planta, incluso las semillas, donde se localizan compuestos tóxicos más concentrados. En las bases de datos científicas, se encontró registro de 193 artrópodos asociados a *R. communis* en diferentes partes del mundo. Esta información se concentró en una base de datos y se analizó de acuerdo con la hipótesis coevolutiva, la cual permite predecir que la mayor riqueza y abundancia de artrópodos fitófagos, se encuentra en el centro de origen de *R. communis*. De esta revisión se desprende que entre las plagas más devastadoras en Asia se encuentran *Achaea janata*, *Spodoptera litura*, *Edwardsiana flavescens*, *Liriomyza trifolii*, *L. sativae*, *Spilosoma obliqua*, *Cogenethes punctiferalis*, *Oxyrhachis taranda* y *Helicoverpa armigera*. En África, las plagas más importantes son *Agrotis ipsilon*, *S. exigua*, *Nezara viridula*, *Trialeurodes ricini* y *Tetranychus urticae*. Entre las plagas de importancia económica que se reportan en Centro y Suramérica, están *Phyllophaga* sp., *Agrietes* sp., *Erinnyis ello*, *N. viridula, Corythucha gossypii*, *Falconia antioquiana* y *S. marima*. Los enemigos naturales de algunas plagas comúnmente reportados fueron *Bacillus thuringiensis*, *B. cereus*, *B. popilliae*, *Trichogramma achaeae*, *T. chilonis*, *T. minutum*, *T. australicum*, *T. dendrolimi*, *T. pretiosum*, *T. evanescens*, *Microplitis rufiventris*, *M. maculipennis*, *M. ophiusae*, *Telenomus remus*, *T. proditor*, *Stethorus siphonulus* y *S. histrio*. Se registra a *Apis mellifera* como el insecto más polinizador de *R. communis*. Los métodos de manejo de plagas contra artrópodos de *R. communis* incluyen control biológico, etológico, mecánico, cultural, génetico y químico.

Palabras clave: Higuerilla, biodiesel, plagas, entomófagos, polinizadores.

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Introduction

The castor-oil plant, *Ricinus communis* L., is an oleaginous plant belonging to the Euphorbiaceae family, which comprises 280 genera. This species has been cultivated for more than 6000 years on the Asian continent, and more recently on the African and American continents (Govaerts *et al*. 2000; Salihu *et al*. 2014). *R. communis* is a non-edible plant, mainly used in chemical, pharmaceutical, and automobile industries, where it has numerous applications (Savy 2005; Barnes *et al*. 2009; Severino *et al*. 2010). All parts of this plant contain lectin ricin – one of the most potent lethal natural poisons known – but is particularly concentrated in the seeds and pods (Audi *et al*. 2005).

In recent years, *R. communis* oil has acquired importance as a biofuel, due to the possibility of its use in producing biodiesel (Baldwin and Cossar 2009; César and Batalha 2010). *R. communis* is distributed in tropical and subtropical regions and is also adaptable to temperate zones (Lima *et al*. 2011). The principal producer countries of *R. communis* seeds are India, China, and Mozambique; whereas the countries with the highest consumption of the products of this plant are Holland, Japan, and Italy (Faostat 2015). India, China, and Brazil contribute approximately 95 % of the world production of seeds (Sailaja *et al*. 2008).

Ricinus communis seeds are outstanding for their high oil content, between 40 and 60 %, compared with sunflower (*Helianthus annuus* L.) seeds with 38 to 48 %, soybean (*Glycine max* (L.) Merr.) between 18 and 19 %, moringa (*Moringa oleifera* L.) with 14 to 24 %, neem [*Azadirachta indica* (Juss)] between 17 and 39 %, and cotton (*Gossypium hirsutum* L.) with 15 to 19 % (Kittock and Williams 1970; Severino *et al*. 2006; Nass *et al*. 2007; Baldwin and Cossar 2009; Martín *et al*. 2010), a characteristic that makes this plant very attractive as a source of biofuel, particularly biodiesel.

The extensive cultivation of varieties and hybrids of *R. communis* under different management practices has made the plant vulnerable to biotic and abiotic factors. *R. communis* plants may lose leaves, seeds and pods for different reasons: damage by pests, diseases, wind, hail, traffic of machinery, and inappropriate use of herbicides and defoliation (Severino *et al*. 2010). Even though a castor-oil plant can recover from severe defoliation, the damage suffered by the leaves may reduce the production. It is estimated that for 1 m^2 of lost leaf area, seed production diminishes by 37.8 g and oil production by 24.4 g (Lakshmamma *et al*. 2009; Lakshmi *et al*. 2010; Severino *et al*. 2010). Continuous sowing of *R. communis* in the same areas, as well as the lack of intercropping has increased the occurrence of pests and diseases. There are reports that more than 100 species of insects in different parts of the world feed on *R. communis* and can cause serious damage (Barteneva 1986; Kolte 1995). In India, for example, insect pests caused losses in seed production from 35 to 50 % (Kolte 1995). Integrated pest management programs are therefore important to prevent losses that can affect the economy of producer-countries.

The present literature reviewed focuses on the phytophagous arthropods associated with *R. communis* in different parts of the world, as well as, their natural enemies and floral visitors. The information was obtained through extense search of scientific literature on these subjects published in the Web of Science database, Ebsco database and Google Scholar, using appropriate key words (e.g. 'insects on *Ricinus communis*' 'arthropods on *Ricinus communis*', 'pests of *Ricinus communis* or castor-oil'); the search was conducted until January 2019. Afterwards, the information collected was analyzed from the perspective of the co-evolutionary hypothesis following the approach of literature review analysis of arthropod herbivory on physic nut (*Jatropha curcas* L.) conducted by Lama *et al*. (2015). Specifically, we set out to answer the following questions regarding the arthropods associated with *R. communis*: (1) What is the diversity of arthropod taxa associated with this plant? (2) In what geographic area does the greatest richness of associated arthropod species occur? (3) What are the parts of the plant most preferred by the herbivorous arthropods? and (4) What mouthpart classes of the arthropods associated with *R. communis* can be identified? According to the co-evolutionary hypothesis, it would be expected to find greater richness of native arthropod species in Asia and Africa, the origin area of *R. communis*, in comparison with those areas where this plant has been introduced or cultivated more recently.

Phytophagous arthropods associated with *R. communis*

Ricinus communis has been considered tolerant and/or resistant to pest attack due to the toxic compounds present in different parts of the plant. Some of the most common compounds found in this plant species are ricin, ricinine, N-demethylricinine, flavonoids, gallic acid, gentisic acid, coumaric acid, syringic acid, cinnamic acid, vanillic acid and rutin, and allergen proteins such as Ric c1 and Ric c3 (Usha Rani *et al*. 2006; Gahukar 2010; Vandenborre *et al*. 2011; Usha Rani and Pratyusha 2014). Some of these are toxic compounds that may even have insecticidal or antifeedant properties against insect pests of other crops (Rossi *et al*. 2012; Amoabeng *et al*. 2014; Dinesh *et al*. 2014). Despite the insecticidal properties of *R. communis*, there are reports of arthropods that feed on several parts of this plant. Ricinine, for example, one of its main alkaloids that has shown insecticidal effect on some insect pests of other plants (Bigi *et al*. 2004; Liu and Li 2006; Rossi *et al*. 2012) does not have any detrimental effect on certain specialist phytophagous insects that are common pests of *R. communis,* such as *Achaea janata* (L., 1758) (Lepidoptera: Noctuidae), *Spodoptera litura* (F., 1775) (Lepidoptera: Noctuidae) and others (Prabhakar *et al*. 2003; Usha Rani and Pratyusha 2014). This is due to the presence of enzymes in the midgut of these insects that are able to degrade toxins and thus breakdown the plants' natural defenses (Yasur *et al*. 2009; Usha Rani and Pratyusha 2014).

The arthropod pests of *R. communis* damage all parts of the plant, including the seeds, where some toxic compounds such as lipases, the alkaloid ricinine (including the protein ricin) and glycosides of ricinoleic, isoricinoleic, stearic and dihydroxystearic acids are even more concentrated (Jena and Gupta 2012). The type of pest and damage varies from place to place; some pests of *R. communis* can be present in different regions. Table 1 presents information published in the literature on arthropods that attack *R. communis*.

According to Table 1, 59 % of the arthropod species feed on foliage, 20 % on roots and seedlings, 17 % on flowers, fruits and seeds, and 5 % on stems and branches. The low percentage of arthropods feeding on seeds and roots can be explained in part by the high concentration of ricinine in these parts of the plant (Salihu *et al*. 2014). To feed on seeds and roots, these arthropods have had to develop highly efficient mechanisms of detoxification (Yasur *et al*. 2009).

Table 1. Order, family and geographical distribution of the phytophagous arthropod species that attack cultivated *Ricinus communis*.

Order	Family	Species	Geographical distribution	References
		Roots and seedlings		
Coleoptera	Curculionidae	*Protostropus* spp.	Africa	Salihu *et al*. (2014)
	Elateridae	*Agriotes* sp.	Costa Rica	Anónimo (1991)
	Scarabeidae	*Amphimallon solstitialis* (Linnaeus, 1758)	Russia	Arkhangel'Skii and Romanova (1930)
		Holotrichia consanguinea Blanchard, 1850	India	Gahukar (2018)
		Phyllophaga sp.	Colombia and Costa Rica	Anónimo (1991); Londoño-Zuluaga (2008)
		Holochelus aequinoctialis (Herbst, 1790) [= *Rhizotrogus aequinoctialis* (Herbst, 1790)]	Russia	Arkhangel'Skii and Romanova (1930)
Diptera	Agromyzidae	*Liriomyza trifolii* (Burgess, 1880)	India	Anjani *et al*. (2007)
Lepidoptera	Noctuidae	*Agrotis ipsilon* (Hüfnagel, 1766)	Colombia and Egypt	Mona *et al*. (2005); Saldarriaga Cardona *et al*. (2011)
		Helicoverpa zea (Boddie, 1850)	USA	Wene (1933)
		Spodoptera frugiperda (J. E. Smith, 1797)	Colombia	Saldarriaga *et al*. (2011)
		Spodoptera marima (Schaus, 1904)	Brazil	Ribeiro and Costa (2008)
	Sphingidae	*Erinnyis ello* (Linnaeus, 1758)	Brazil	Ribeiro and Costa (2008)
Orthoptera	Gryllidae	*Brachytrupes* spp.	Africa	Salihu *et al*. (2014)
	Pyrgomorphidae	*Chrotogonus* spp.	Africa	Salihu *et al*. (2014)
		Zonocerus variegatus (Linnaeus, 1758)	Africa	Salihu *et al*. (2014)
Isoptera	Termitidae	*Odontotermes obesus* (Rambur, 1842)	India	Gahukar (2018)
		Leaves		
Coleoptera	Curculionidae	*Naupactus glaucus* Perty, 1832 [= *Pantomorus glaucus* (Perty, 1830)]	Brazil	Cavalcante *et al*. (1974)
Diptera	Agromyzidae	*Liriomyza sativae* Blanchard, 1938	China	Zhang *et al*. (2006)
		Liriomyza subpusilla Frost, 1943	USA	Wene (1933); Parkman *et al*. (1989)
		Liriomyza trifolii (Burgess, 1880)	India	Galande *et al*. (2005)
Hemiptera	Aleyrodidae	*Bemisia tabaci* (Gennadius, 1889)	Costa Rica and Africa	Anónimo (1991); Salihu *et al*. (2014)
		Trialeurodes ricini (Misra, 1924) (= *Trialeurodes rara* Singh, 1931)	India and Egypt	Idriss *et al*. (1997); Sarma *et al*. (2005); Abdullah and Martin (2007); Raghavaiah (2011)
	Aphrophoridae	*Ptyelus grossus* (Fabricius, 1781)	Uganda	Darling (1946)
	Cicadellidae	*Amrasca* (*Amrasca*) *biguttula* (Ishida, 1913) [= *Amrasca biguttula biguttula* (Ishida, 1912)]	India	Sharma and Singh (2002); Raghavaiah (2011)
		Agallia sp.	Spain	Durán *et al*. (2010)
		Edwardsiana flavescens (Fabricius, 1794) [= *Empoasca flavescens* ((Fabricius, 1794)]	India	Jayaraj (1964); Sarma *et al*. (2005); Lakshmi *et al*. (2005); Jyothsna *et al*. (2009)
		Empoasca (*Empoasca*) *solana* Delong, 1931 (= *Empoasca solana* Delong, 1931)	USA	Wene (1933)
		Empoasca sp.	Costa Rica	Anónimo (1991)
		Empoasca sp.	Africa	Salihu *et al*. (2014)
		Jacobiasca furcostylus (Ramakrishnan y Menon, 1972)	India	Parmar *et al*. (2006)
	Miridae	*Falconia antioquiana* Carvalho, 1987	Colombia	Saldarriaga Cardona *et al*. (2011)
		Polymerus cognatus (Fieber, 1858) (= *Poeciloscytus cognatus* Fieber, 1858)	Russia	Arkhangel'Skii and Romanova (1930)
	Pentatomidae	*Acrosternum pallidoconspersum* (Stål, 1858)	Egypt	Jannone (1952)
		Nezara viridula (Linnaeus, 1758)	Costa Rica and Egypt	Jannone (1952); Anónimo (1991)
	Pseudococcidae	*Paracoccus marginatus* Williams and Granara de Willink, 1992	Cuba	Martínez *et al*. (2005)
	Tingidae	*Corythucha gossypii* (Fabricius, 1794)	USA, Colombia, Mexico, and Cuba	Miller and Nagamine (2005); Londoño-Zuluaga (2008); Saldarriaga Cardona *et al*. (2011), López-Guillén *et al*. (2012)
Lepidoptera	Arctiidae	*Amsacta moorei* Butler, 1876	India	Sarma *et al*. (2005)
		Amsacta albistriga Walker, 1864	India	Sarma *et al*. (2005)
		Pericallia ricini (Fabricius, 1775)	India	Mathur *et al*. (1994); Neelanarayanan and Indira (2010)
		Spilosoma obliqua Walker, 1855	India and Pakistan	Singh and Grewal (1982); Khattak *et al*. (1991); Sarma *et al*. (2005)

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	Dalceridae	*Anacraga citrinopsis* Dyar, 1927	Brazil	Lourenção *et al*. (1989)
	Limacodidae	*Parasa lepida* Cramer, 1799	India	Raghavaiah (2011)
	Lymantriidae	*Dasychira* sp.	Africa	Salihu *et al*. (2014)
		Euproctis fraterna Moore, 1883	India	Paul *et al*. (2000); Suganthy (2010)
	Noctuidae	*Achaea janata* (Linnaeus, 1758)	India, USA, and China	Hua (1984); Delaya *et al*. (1985); Basappa and Lingappa (2001); Mau and Kessing (2007)
		Helicoverpa armigera (Hübner, 1803-1808)	India and USA	Wene (1933); Ribeiro and Costa (2008)
		Spodoptera cosmioides (Walker, 1858)	Brazil	Bavaresco *et al*. (2003)
		Spodoptera exigua (Hübner, 1808)	Egypt	Ribeiro and Costa (2008)
		Spodoptera litura (Fabricius, 1775)	India and Pakistan	Lohar *et al*. (1997); Usha Rani and Rajasekharreddy (2009)
		Spodoptera ornithogalli (Guenée, 1852) [= *Spodoptera marima* (Schaus, 1904)]	Brazil	Ribeiro and Costa (2008)
		Spodoptera sp.	Costa Rica	Anónimo (1991)
	Nymphalidae	*Ariadne merione* Cramer, 1779 (= *Ergolis merione* Cramer, 1779)	India	Ghosh (1914); Sarma *et al*. (2005)
	Saturniidae	*Samia ricini* (Drury, 1773)	Egypt, India, and Brazil	El-Shaarawy *et al*. (1975); Negreiros *et al*. (1998)
		Rothschildia jacobaeae Walker, 1855	Brazil	Ribeiro and Costa (2008)
Orthoptera	Acrididae	*Chrotogonus* (*Chrotogonus*) *trachypterus robertsi* Kirby & W. F., 1914 (= *Chrotogonus robertsi* Kirby & W. F., 1914)	India	Sarma *et al*. (2005)
Thysanoptera	Thripidae	*Retithrips syriacus* (Mayet, 1890)	India	Sarma *et al*. (2005)
		Scirtothrips dorsalis Hood, 1919	India	Patel *et al*. (2009)
		Zaniothrips ricini Bhatti, 1967	India	Daniel *et al*. (1983)
Acarina	Tetranychidae	*Eutetranychus orientalis* (Klein, 1936)	India	Ahuja (1994)
		Eutetranychus sp.	India	Raghavaiah (2011)
		Tetranychus piercei McGregor, 1950	China	Lui and Lui (1986)
		Tetranychus urticae Koch, 1836 [= *Tetranychus telarius* (Linnaeus, 1758)]	Morocco and India	Cangardel (1954); Rajasekhar *et al*. (1999); Raghavaiah (2011)
	Tarsonemidae	*Polyphagotarsonemus latus* (Banks, 1904)	Belgium	Heungens and Degheele (1986)
		Stems and branches		
Coleoptera	Buprestidae	*Sphenoptera* sp.	Africa	Salihu *et al*. (2014)
	Tenebrionidae	*Blapstinus* sp.	USA	De Ong (1918)
Hemiptera	Membracidae	*Oxyrhachis taranda* (Fabricius, 1798)	India	Ali *et al*. (2006)
Lepidoptera	Cossidae	*Strigocossus capensis* (Walker, 1856) [= *Xyleutes capensis* (Walker, 1856)]	Africa	Salihu *et al*. (2014)
		Flowers, fruits and seeds		
Coleoptera	Anobiidae	*Lasioderma serricorne* (Fabricius, 1792)	India and Africa	Hussain and Khan (1966); Salihu *et al*. (2014)
	Tenebrionidae	*Tribolium castaneum* (Herbst, 1797)	Africa	Salihu *et al*. (2014)
Hemiptera	Cicadellidae	*Empoasca* sp.	Costa Rica	Anónimo (1991)
	Miridae	*Eurystylus* sp.	Africa	Salihu *et al*. (2014)
		Helopeltis sp.	Africa	Salihu *et al*. (2014)
	Pentatomidae	*Nezara viridula* (Linnaeus, 1758)	Costa Rica, and USA	Anónimo (1991); Golden and Follett (2006)
	Scutelleridae	*Calidea* sp.	Africa	Salihu *et al*. (2014)
Lepidoptera	Crambidae	*Conogethes punctiferalis* (Guenée, 1854) [= *Dichocrocis punctiferalis* (Guenée, 1854)]	India and Australia	Anonymous (1913); Sharma *et al*. (1995); Jyothsna *et al*. (2009); Patel and Patel (2009); Hedge *et al*. (2009)
	Noctuidae	*Achaea janata* (Linnaeus, 1758)	India, USA, and China	Hua (1984); Delaya *et al*. (1985); Basappa and Lingappa (2001); Mau and Kessing (2007)
		Heliothis sp.	Costa Rica	Anónimo (1991)
		Helicoverpa armigera (Hübner, 1803-1808)	India, and USA	Wene (1933); Geetha *et al*. (2003); Satyanarayana and Sing (2003)
		Spodoptera sp.	Costa Rica	Anónimo (1991)
	Pyralidae	*Cadra cautella* (Walker, 1863) [= *Ephestia cautella* (Walker, 1863)]	Africa	Salihu *et al*. (2014)
	Tortricidae	*Thaumatotibia leucotreta* (Meyrick, 1913) (= *Cryptophlebia leucotreta* Meyrick, 1913)	Africa	Salihu *et al*. (2014)

A total of 76 species of phytophagous arthropods associated to cultivated plants of *R. communis* is found worldwide (Table 1). Before the present literature review, the report was of 60 species (Raoof *et al*. 2003). The arthropods reported in Table 1 belong to eight orders and 38 families; 40 % of these species belong to Lepidoptera, 27 % to Hemiptera, 14 % to Coleoptera and 19 % to other orders. The species that belong to Lepidoptera, Hemiptera and Coleoptera represent 81 % of the total. These phytophagous arthropods are distributed geographically in Asia (39 %), America (34 %), Africa (25 %) and Europe (2 %). As it was supposed, it was not uncommon to find that the greatest richness of arthropods associated to *R. communis* occurred in Asia and Africa, continents considered as the center of origin of this plant (Govaerts *et al*. 2000). 63 % of the species had mandibulate mouthparts (Lepidoptera, Coleoptera, Orthoptera, Isoptera and Diptera) and 37 % were piercing-and-sucking mouthpart classes (Hemiptera, Thysanoptera and Acarina).

Of the pests listed in Table 1, the castor semilooper *A. janata*, the tobacco caterpillar *S. litura*, the green leafhopper *Edwardsiana flavescens* (F., 1794) [= *Empoasca flavescens* (F., 1794)] (Hemiptera: Cicadellidae), the serpentine leafminer *Liriomyza trifolii* Burgess, 1880, the vegetable leafminer *L. sativae* Blanchard, 1938 (Diptera: Agromyzidae), the Bihar hairy caterpillar *Spilosoma obliqua* Walker, 1855 (Lepidoptera: Arctiidae), the shoot and capsule borer *Conogethes punctiferalis* (Guenée, 1854) [= *Dichocrocis punctiferalis* (Guenée, 1854)] (Lepidoptera: Crambidae), the cowbug *Oxyrhachis taranda* (F., 1798) (Hemiptera: Membracidae), and the cotton bullworm *Helicoverpa armigera* (Hübner, 1803-1808) (Lepidoptera: Noctuidae), among others, are the most devastating pests in Asia. In Africa, the black cutworm *Agrotis ipsilon* (Hüfnagel, 1776), the armyworm *S. exigua* (Hübner, 1808) (Lepidoptera: Noctuidae), the stink bug *Nezara viridula* (L., 1758) (Hemiptera: Pentatomidae), the castor bean whitefly *Trialeurodes ricini* (Misra, 1924) (= *Trialeurodes rara* Singh, 1931) (Hemiptera: Aleyrodidae), the red spider mite *Tetranychus urticae* Koch, 1836 [= *Tetranychus telarius* (Linnaeus, 1758)] (Acarina: Tetranychidae), among others, are mentioned as the most important. In Central and South-America, the white grub *Phyllophaga* sp. (Coleoptera: Scarabaeidae), *Agrietes* sp., *Erinnyis ello* (F., 1794) (Lepidoptera: Sphingidae), *N. viridula,* the cotton lace bug *Corythucha gossypii* (Fabricius, 1794) (Hemiptera: Tingidae), the sucking bug *Falconia antioquiana* Carvalho, 1987 (Hemiptera: Miridae), *S. marima* (Schaus, 1904) (Lepidoptera: Noctuidae) and others, are reported as pests of economic importance for *R. communis* (Varón *et al*. 2010; Saldarriaga Cardona *et al*. 2011; López-Guillén *et al*. 2012).

The principal pests in Brazil are *N. viridula*, the leafhopper *Empoasca* spp., some defoliator larvae including *S. frugiperda* Smith (Lepidoptera: Noctuidae), *A. janata*, and *A. ipsilon*, and mites such as *T. urticae*, and *T. ludeni* Zacher, 1913 (Acarina: Tetranychidae) (Soares *et al*. 2001; Ribeiro and Costa 2008). In Colombia, *C. gossypii* is mentioned as the pest of greatest economic importance in *R. communis* crops (Varón *et al*. 2010). In Mexico, *C. gossypii, N. viridula, and Tetranychus* spp. are reported as the main potential pests of *R. communis* (López-Guillén *et al*. 2012) (Table 1).

Phytophagous arthropods found on noncultivated *R. communis*

There are reports of phytophagous insects mostly found in noncultivated *R. communis* plants, isolated plants, as well as, in more or less clustered plants or plants growing in urban and suburban areas, and in disturbed landscapes of Egypt, India, Spain, Uganda, USA and other countries (Oshaibah *et al*. 1986; Singh *et al*. 1991; Jacob *et al*. 2000; Pons *et al*. 2002; Ylla *et al*. 2008; Boland 2016; Egonyu *et al*. 2017).

Table 2 presents a total of 20 species of phytophagous arthropods associated with non-cultivated plants of *R. communis* in the world. These species belong to five orders and 16 families. 60 % of the arthropod species belong to Lepidoptera (30 %) and Hemiptera (30 %), while 40% belong to Coleoptera (20 %) and other orders (20 %). The species that belong to Lepidoptera, Hemiptera and Coleoptera represent 80 % of the total. 59 % of the arthropod species registered in Table 2 are distributed geographically in Asia (32 %) and Africa (27 %), while 41 % are registered in America (32 %) and Europe (9 %). 60 % of the species of arthropods are mandibulate mouthpart (Lepidoptera, Coleoptera, and Orthoptera) and 40 % are piercing-and-sucking mouthpart (Hemiptera and Acarina).

Such insects were observed feeding on leaves of *R. communis* plants, and even though some species have been reported as pests of *R. communis* in other countries, most of them cause no considerable damage. However, they have the potential of becoming pests of *R. communis* if it is cultivated as a monoculture or, *R. communis* could be a plant host for important pests as the invasive ambrosia beetle *Euwallacea* sp. (Coleoptera: Curculionidae) (Boland 2016; Egonyu *et al*. 2017). Among these potential pests are insect and mite species of various families of Lepidoptera, Hemiptera, Orthoptera, and others (Table 2).

Pollinator insects and floral visitors in *R. communis*

Ricinus communis is a monoecious cross-pollinating plant, cultivated as a hybrid in India, Brazil, China, and other countries because they produce better yields than pure lines or varieties (Moll *et al*. 1962; Birchler *et al*. 2003; Reif *et al*. 2007). Several studies demonstrate that certain species of pollinator insects may improve seed production of *R. communis*. For example, it is mentioned that *Apis mellifera* (L., 1758) (Hymenoptera: Apidae) contributes to increasing *R. communis* crop productivity by incrementing fruit numbers as well as oil content in seeds (Freitas and Cruz 2010).

Among the pollinator insects of *R. communis, A. mellifera* is recorded as the main pollinating insect. It is also mentioned that this insect feeds on the nectar produced by the plant's extrafloral nectar glands (Rizzardo *et al*. 2012; Waters *et al*. 2014). *A. mellifera* is the principal pollinating insect of *R. communis,* and laboratory work has demonstrated that the pollen of this plant reduces bee survival (Junior *et al*. 2011). According to these studies, expansion of *R. communis* as a crop in the semiarid region of Brazil for biodiesel production represents a risk for the native and domestic bees used for honey production.

As shown in Table 3, a total of 36 species of pollinator insects and floral visitors of non-cultivated plants of *R. communis* is found in the world. These species belong to four orders and 16 families. 25 % of the species belong

Table 2. Order, family and geographical distribution of the phytophagous arthropod species found on non-cultivated *Ricinus communis.*

Order	Family	Species	Geographical distribution	References
Leaves				
Coleoptera	Bostrichidae	*Prostephanus truncatus* (Horn, 1878)	Mexico	Bourne-Murrieta *et al*. (2014)
	Chrysomelidae	*Diabrotica graminea* Baly, 1886	Puerto Rico	Woloott (1917)
	Scarabaeidae	*Lepadoretus sinicus* Burmeister, 1855 (= *Adoretus sinicus* Burmeister, 1855)	USA	McQuate y Jameson (2011)
	Scolytidae	*Euwallacea* sp.	Uganda and USA	Boland (2016); Egonyu *et al*. (2017)
Hemiptera	Aleyrodidae	*Aleurodicus dispersus* Russell, 1965	Cape Verde	Monteiro *et al*. (2005)
	Cicadellidae	*Amrasca* (*Amrasca*) *biguttula* (Ishida, 1913) [= *Amrasca devastans* (Distant, 1918)]	India	Jacob *et al*. (2000)
		Empoasca (*Empoasca*) *kerri* Singh-Pruthi, 1940 (= *Empoasca kerri* Pruthi, 1940)	India	Singh *et al*. (1991); Jacob *et al*. (2000)
		Empoasca (*Empoasca*) *motti* Singh-Pruthi, 1940 (= *Empoasca motti* Singh-Pruthi, 1940)	India	Jacob *et al*. (2000)
	Flatidae	*Metcalfa pruinosa* (Say, 1830)	Spain	Pons *et al*. (2002)
	Miridae	*Apolygus lucorum* (Meyer-Dür, 1843)	China	Lu *et al*. (2010)
Lepidoptera	Arctiidae	*Amsacta moorei* Butler, 1876	India	Singh *et al*. (1989)
	Cosmopterigidae	*Pyroderces rileyi* (Walsingham, 1882) (= *Sathrobrota rileyi* Walsingham, 1882)	Egypt	Oshaibah *et al*. (1986)
	Lymantriidae	*Euproctis lunata* Walker, 1855	Bangladesh	Islam *et al*. (1988)
	Noctuidae	*Agrotis ipsilon* (Hüfnagel, 1766)	Egypt	Younis (1992)
	Pyralidae	*Phycita diaphana* (Staudinger, 1870)	Spain	Huertas Dionisio (2002); Ylla *et al*. (2008)
	Tortricidae	*Thaumatotibia leucotreta* (Meyrick, 1913) (= *Cryptophlebia leucotreta* Meyrick, 1913)	South Africa	Kirkman and Moore (2007)
Orthoptera	Acrididae	*Melanoplus differentialis* (Thomas, 1865)	USA	Spain (1940)
Acarina	Tetranychidae	*Eutetranychus banksi* (McGregor, 1914)	USA	McGregor (1914)
		Eutetranychus orientalis (Klein, 1936)	Palestine and Egypt	Klein (1936)
		Tetranychus gloveri Banks, 1900 (= *Tetranychus quinquenychus* McGregor, 1914)	USA	McGregor (1914)

to Lepidoptera (19 %) and Hemiptera (6 %), while 75 % belong to Hymenoptera (67 %) and Diptera (8 %). 55 % of the arthropod species registered in Table 3 are distributed geographically in Asia (33 %) and Africa (22 %), while 45 % are registered in America; no records were found for Europe.

In Mexico, Cameroon, USA, India, and Brazil, entomophagous Hymenoptera, as well as several species of Lepidoptera, Diptera, and Hemiptera have been reported to feed on nectaries and flowers of *R. communis*; however, only *A. mellifera* has been reported as a pollinator. Therefore, it is necessary to carry out studies on pollination and floral ecology in order to determine if there are other insect pollinators of *R. communis* that should be protected or may be used to increase crop yield (Table 3).

Some pests can affect pollinators through herbivory. In the case of *R. communis*, Wäckers *et al*. (2001) showed that plants damaged by larvae of *Spodoptera littoralis* (Boisd., 1833) (Lepidoptera: Noctuidae) increased the total amount of nectar produced by extrafloral nectaries compared to undamaged plants. De Sibio and Rossi (2016) found a similar result for the herbivory of *S. frugiperda* on *R. communis*. The secretion of carbohydrates through extrafloral nectaries is considered an indirect strategy of plant defense because it serves to attract parasitoids and predators (Heil 2008). Unlike floral nectaries, extrafloral nectaries do not participate in pollination, however, in plants pollinated by insects, extrafloral nectaries can negatively affect the effectiveness of pollination by distracting pollinators away from floral nectaries or when the ants that are attracted by the nectar attack the floral visitors (Wäckers *et al*. 2001; Turlings and Wäckers 2004).

Natural enemies of the pests of *R. communis*

Among the natural enemies of the key pests of cultivated *R. communis,* there are parasitoids, predators, and entomopathogens such as fungi, bacteria, nematodes, and viruses, which are used as biological control agents or have been found parasitizing, depredating, or naturally infecting some pests of the crop. An extensive list of natural enemies of phytophagous arthropods of *R. communis* grouped by taxa with information of their host or prey and geographical distribution is shown in Table 4; as it can appreciate in this table, the most commonly reported natural enemies in countries like India, Brazil, China, and USA, are *Bacillus* spp., *Trichogramma* spp., *Microplitis* spp., *Telenomus* spp., *Stethorus* spp., and other species attacking pests such as *A. janata, S. litura, Anacraga citrinopsis* Dyar, 1927*, S. obliqua, Phyllophaga* sp., *Eutetranychus banksi* (McGregor, 1914), *Tetranychus piercei* McGregor, 1950*, Zaniothrips ricini* Bhatti, 1967*,* and other species. Table 4 shows a total of 61 natural enemies of phytophagous insects of *R. communis*. Three species are bacteria belonging to the same genus; four species are nematodes of different genera; two species are fungi of different genera; two reports are viruses; 36 species are parasitoids of eight families of Hymenoptera and one family of Diptera; and 14 species are predators of six different families and order 74 % of the species is distributed geographically in Asia, 24 % in America, 2 % in Africa and 0 % in Europe.

An example of natural enemies of pest of *R. communis* is presented by Basappa (2009). According to this author, parasitoids, insect predators, spiders, insectivorous birds and some microbial organisms are important natural enemies of the pest complex of *R. communis* ecosystem in India. In the case of *A. janata*, *Trichogramma chilonis* Ishii, 1941, *Trichogramma achaeae* Nagaraja and Nagarkatti, 1970, *Telenomus* sp. and *Trissolcus* sp. were recorded from eggs; *Microplitis maculipennis* (Szepligeti, 1900), *Euplectrus*

Table 3. Order, family and geographical distribution of the pollinators and floral visitors, reported on *Ricinus communis* plants.

Order	Family	Species	Geographical distribution	References
Diptera	Muscidae	*Musca domestica* Linnaeus, 1758	Cameroon	Douka and Tchuenguem (2014)
	Richardiidae	*Sepsisoma* sp.	Brazil	Souza-Silva *et al*. (2001)
	Syrphidae	*Ischiodon scutellaris* (Fabricius, 1805)	India	Navatha and Sreedevi (2012)
Hemiptera	Coreidae	*Anoplocnemis curvipes* (Fabricius, 1871)	Cameroon	Douka and Tchuenguem (2014)
	Pentatomidae	*Nezara viridula* Linnaeus, 1758	India	Navatha and Sreedevi (2012)
Lepidoptera	Nymphalidae	*Acraea acerata* (Hewitson, 1874)	Cameroon	Douka and Tchuenguem (2014)
		Acraea terpsicore (Linnaeus, 1758)	India	Navatha and Sreedevi (2012)
		Hypolimnas misippus (Linnaeus, 1764)	India	Navatha and Sreedevi (2012)
	Pieridae	*Catopsilia florella* (Fabricius, 1775)	Cameroon	Douka and Tchuenguem (2014)
		Eurema blanda (Boisduval, 1836)	India	Navatha and Sreedevi (2012)
		Eurema sp.	Cameroon	Douka and Tchuenguem (2014)
		Pieris brassicae (Linnaeus, 1758)	India	Navatha and Sreedevi (2012)
Hymenoptera	Apidae	*Apis mellifera* Linnaeus, 1758	Brazil	Freitas *et al*. (2009); Freitas and Cruz (2010)
		Apis florea Fabricius, 1973	India	Navatha and Sreedevi (2012)
		Ceratina sp.	India	Navatha and Sreedevi (2012)
		Scaptotrigona sp.	Brazil	Freitas *et al*. (2009)
		Trigona sp.	India	Navatha and Sreedevi (2012)
		Xylocopa fenestrata (Fabricius, 1798)	India	Navatha and Sreedevi (2012)
	Braconidae	*Bracon* spp.	Mexico	Álvarez and Reyes (1987)
		Ephiaulax sp.	Mexico	Álvarez and Reyes (1987)
	Chalcididae	*Conura igneoides* Kirby, 1883 [= *Spilochalcis igneoides* (Kirby, 1883)]	Mexico	Álvarez and Reyes (1987)
		Conura maria Riley, 1870 [= *Spilochalcis mariae* (Riley, 1872)]	Mexico	Álvarez and Reyes (1987)
	Eurytomidae	*Neorileya* sp.	Mexico	Álvarez and Reyes (1987)
	Formicidae	*Camponotus compressus* (Fabricius, 1787)	India	Navatha and Sreedevi (2012)
		Linepithema humile (Mayr, 1868)	USA	Line *et al*. (2013)
		Polyrachis sp.	Cameroon	Douka and Tchuenguem (2014)
	Halictidae	*Halictus* sp.	India	Navatha and Sreedevi (2012)
	Sphecidae	*Liris* sp.	Mexico	Álvarez and Reyes (1987)
		Sceliphron assimile (Dahlbom, 1843)	Mexico	Álvarez and Reyes (1987)
		Tachysphex sp.	Mexico	Álvarez and Reyes (1987)
		Tachytes sp.	Mexico	Álvarez and Reyes (1987)
		Trypoxilon sp.	Mexico	Álvarez and Reyes (1987)
	Torymidae	*Torymus capillaceus* (Huber, 1927)	Mexico	Álvarez and Reyes (1987)
	Vespidae	*Synagris cornuta* (Linnaeus, 1758)	Cameroon	Douka and Tchuenguem (2014)
		Delta sp.	Cameroon	Douka and Tchuenguem (2014)
		Polistes sp.	Mexico	Álvarez and Reyes (1987)

Table 4. Natural enemies of phytophagous arthropods of *Ricinus communis*.

Species	Host and/or prey	Geographical distribution	References
Entomopathogens			
Bacteria			
Bacillus thuringiensis var. *kurstaki* (Berliner, 1915)	Larvae of *Achaea janata*	India	Vimala Devi and Sudhakar (2006)
Bacillus cereus (Manson, Pollock & Tridgell, 1954)	Larvae of *Achaea janata*	India	Kattegoudar *et al*. (1994)
Bacillus popilliae Dutky, 1940	Larvae of *Phyllophaga* sp.	Colombia	Saldarriaga Cardona *et al*. (2011)
Nematodes			
Hexamermis dactylocercus Poinar and Linares, 1985	Larvae of *Amsacta albistriga*	India	Prabhakar *et al*. (2010)
Steinernema carpocapsae (Weiser, 1955)	Larvae of *Spodoptera litura*	India	Raveendranath *et al*. (2008)
Heterorhabditis indica Poinar, Karunaka y David, 1992	Larvae of *Spodoptera litura*	India	Raveendranath *et al*. (2008)
Mermis sp.	Larvae of *Achaea janata*	India	Sujatha *et al*. (2011)
Fungi			
Metarhizium rileyi (Farl.) Kepler, S.A.Rehner & Humber, 2014 [= *Nomuraea rileyi* (Farlow) Samson, 1974]	Larvae of *Spodoptera litura*	India and USA	Mau and Kessing (2007)
Beauveria bassiana (Balsamo) Vuillemin, 1912	Larvae of *Achaea janata* and *Cogenethes punctiferalis*	India	Duraimurugan *et al*. (2015)
Virus			
Nucleopolyhedrovirus	Larvae of *Spodoptera litura*	India	Basappa (2009)
Granulovirus	Larvae of *Achaea janata* and *Spodoptera litura*	India	Naveen Kumar *et al*. (2013)
Parasitoids			
INSECTA			
<u>Hymenoptera</u>			
Aphelinidae			
Encarsia formosa Gahan, 1924	Nymphs of *Trialeurodes ricini*	China	Wang *et al*. (2016)
Braconidae			
Habrobracon hebetor (Say, 1836)	Larvae of *Cogenethes punctiferalis*	India	Basappa (2003)
Apanteles hyposidrae Wilkinson, 1928	Larvae of *Achaea janata*	India	Basappa (2009)
Apanteles ricini Bhatnagar, 1948	Larvae of *Cogenethes punctiferalis*	India	Basappa (2003)
Cotesia flavipes (Cameron, 1891) [*Apanteles flavipes* (Cameron, 1891)]	Larvae of *Spilosoma obliqua* and *Spodoptera litura*	India	Yadav *et al*. (2010); Basappa (2009)
Glyptapanteles dalosoma de Santis, 1987	Larvae of *Anacraga citrinopsis*	Brazil	Lourenção *et al*. (1989)
Microplitis (= *Microgaster*) *rufiventris* Kokujev, 1914	Larvae of *Spodoptera litoralis*	Egypt	Shalaby *et al*. (1988)
Microplitis maculipennis (Szepligeti, 1900) (= *Microplitis ophiusae* Aiyar, 1921)	Larvae of *Achaea janata*	India	Suganthy (2010); Naik *et al*. (2010)
Chalcididae			
Brachymeria euploeae (Westwood, 1837)	Pupae of *Cogenethes punctiferalis*	India	Sujatha *et al*. (2011)
Eulophidae			
Ceranisus menes (Walker, 1839)	2º instar nymph of *Zaniothrips ricini*	India	Daniel *et al*. (1983)
Euplectrus maternus Bhatnagar, 1952	Larave of *Achaea janata*	India	Basappa (2009)
Tetrastichus howardi (Olliff, 1893) (= *Tetrastichus ayyari* Rohwer, 1921)	Pupae of *Spodoptera litura* and *Achaea janata*	India	Basappa (2009)
Trichospilus pupivorus Ferrière, 1930	Pupae of *Spodoptera litura* and *Achaea janata*	India	Basappa (2009)
Trichogrammatidae			
Trichogramma achaeae Nagaraja and Nagarkatti, 1970	Eggs of *Achaea janata*	India	Basappa (2009)
Trichogramma chilonis Ishii, 1941	Eggs of *Achaea janata* and *Spodoptera litura*	India	Singh *et al*. (2008); Suganthy (2010); Naik *et al*. (2010)
Trichogramma minutum Riley, 1879	Eggs of *Achaea janata*	USA	Mau and Kessing (2007)
Trichogramma australicum Girault, 1912	Eggs of *Achaea janata*	China	Hua (1984)
Trichogramma dendrolimi Matsumura, 1926	Eggs of *Achaea janata*	China	Hua (1984)
Trichogramma pretiosum Riley, 1879	Eggs of *S. cosmioides*	Brazil	Cabezas *et al*. (2013)
Trichogramma evanescens Westwood, 1833	Eggs of *Achaea janata*	India	Basappa (2009)

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Scelionidae			
Telenomus remus Nixon, 1937	Eggs of *Spodoptera litura*, *Spodoptera cosmioides* and *Spodoptera frugiperda*	India	Satyanarayana *et al*. (2005); Pomari *et al*. (2013)
Telenomus proditor Nixon, 1937	Eggs of Lepidoptera	USA	Mau and Kessing (2007)
Telenomus sp.	Eggs of *Achaea janata*	India	Basappa (2009)
Trissolcus sp.	Eggs of *Achaea janata*	India	Basappa (2009)
Vespidae			
Polistes sp.	Larvae of *Phyllophaga* sp., *Agrotis* sp. and *Spodoptera* spp.	Costa Rica	Anónimo (1991)
Ichneumonidae			
Campoletis chlorideae Uchida, 1957	Larvae of *Spodoptera litura*	India	Satyanarayana *et al*. (2005)
Charops obtusus Morley, 1913	Larvae of *Spilosoma obliqua* and *Achaea janata*	India	Basappa (2009)
Hyposoter exiguae (Viereck,1912)	Larvae of *Achaea janata*	USA	Mau and Kessing (2007)
Diadegma ricini Row & Kurian,1950	Larvae of *Cogenethes punctiferalis*	India	Basappa (2003)
Theronia sp.	Larvae of *Cogenethes punctiferalis*	India	Basappa (2003)
Isdromas monterai (Costa Lima, 1948)	Larvae of *Anacraga citrinopsis*	Brazil	Lourenção *et al*. (1989)
Tachinidae			
Palexorista parachrysops Bezzi, 1925	Larvae of *Cogenethes punctiferalis*	India	Kalra (1984)
Eucelatoria armigera (Coquillett, 1889)	Larvae and pupae of *Achaea janata*	USA	Mau and Kessing (2007)
Chaetogaedia monticola (Bigot, 1887)	Larvae and pupae of *Achaea janata*	USA	Mau and Kessing (2007)
Predators			
<u>Coleoptera</u>			
Carabidae			
Calosoma sp.	Larvae of *Phyllophaga* sp., *Agrotis* sp. and *Spodoptera* spp.	Costa Rica	Anónimo (1991)
Coccinellidae			
Cheilomenes sexmaculata (Fabricius, 1781)	Eggs larvae of *Achaea janata* and *Spodoptera litura*	India	Basappa (2009)
Micraspis cardoni (Weise, 1892)	*Zaniothrips ricini*	India	Daniel *et al*. (1983)
Scymnus sp.	*Eutetranychus orientalis*	Palestine and Egypt	Klein (1936)
Stethorus sp.	*Eutetranychus banksi*	USA	McGregor (1914)
Stethorus siphonulus Kapur, 1948	*Tetranychus piercei*	China	Lui and Lui (1986)
Stethorus histrio Chazeau, 1974	*Tetranychus urticae*	Chile	Aguilera (1987)
<u>Hemiptera</u>			
Pentatomidae			
Eocanthecona furcellata (Wolff, 1811)	*Achaea janata*	India	Rao (1977); Usha Rani (2009)
Reduviidae			
Rhynocoris kumarii Ambrose and Livingstone, 1986	Eggs larvae of *Achaea janata* and *Spodoptera litura*	India	Basappa (2009)
<u>Thysanoptera</u>			
Aeolothripidae			
Franklinothrips megalops (Trybom, 1912)	*Zaniothrips ricini*	India	Daniel *et al*. (1983)
Mymarothrips garuda Ramakrishna and Margabandhu, 1931	*Zaniothrips ricini*	India	Daniel *et al*. (1983)
<u>Neuroptera</u>			
Chrysopidae			
Chrysoperla carnea (Stephens, 1836)	*Tetranychus urticae*	India	Rajasekhar *et al*. (1999)
Chrysoperla sp.	Eggs and larvae of *Achaea janata* and *Spodoptera litura*	India	Basappa (2009)
<u>Mantodea</u>			
Mantidae			
Haldwania lilliputana Beier, 1930	*Zaniothrips ricini*	India	Daniel *et al*. (1983)
ARACHNIDA / Acari			
Phytoseiidae			
Sciulus sp.	*Eutetranychus banksi*	USA	McGregor (1914)

maternus Bhatnagar, 1952, *Rhogas* spp. and *Apanteles hyposidrae* Wilkinson, 1928 were found among larval parasitoids; and pupae were found to be parasitised by *Tetrastichus howardi* (Olliff, 1893) (= *Tetrastichus ayyari* Rohwer, 1921) and *Phorocera* sp. Among insect predators of *A. janata*, *Chrysoperla* sp. and *Cheilomenes sexmaculata* (Fabricius, 1781) were found feeding on the eggs and neonate larvae; other general insect predators like mantids, paper wasps, and sphecid digger wasps were also found predating on larvae; spiders like green lynx spiders, jumping spiders and crab spiders were found feeding on early instar larvae. Among the entomopathogens of *A. janata*, *Spodoptera litura* nucleopolyhedrovirus and granulosis virus were isolated from dead larvae and the fungi *Metarhizium rileyi* (Farl.) Kepler, S.A.Rehner & Humber, 2014 [= *Nomuraea rileyi* (Farlow) Samson, 1974] and *Beauveria bassiana* (Balsamo) Vuillemin were found infecting larvae (Basappa 2009). Many other examples of natural enemies of pests of *R. communis* are shown in Table 4.

Pest management of phytophagous arthropods in *R. communis*

Pest management methods used to control the principal arthropod pests of *R. communis* include cultural, genetic, ethological, biological, and chemical control.

Cultural control is the use of agronomical practices designed to reduce the presence of pests in crops of *R. communis.* Intercropping is a type of cultural control recommended to diminish the damage caused by insect pests in *R. communis.* Srinivasa Rao *et al*. (2012) found that plants such as *Cyamopsis tetragonoloba* (L.) Taub., 1891, *Vigna unguiculata* (L.) Walp., 1845, *Vigna mungo* (L.) Hepper, 1956, and *Arachis hypogaea* L., 1753, intercropped with *R. communis* in a 1:2 proportion, decreased the incidence of insect pests such as *A. janata, E. flavescens,* and *C. punctiferalis*. Moreover, a more considerable presence of natural enemies of these pests was observed in these intercropping systems. Patel and Patel (2009) recommended intercropping *R. communis* with *Vigna radiata* (L.) Wilczek, 1954, *Sesamum indicum* L., 1753, *Vigna aconitifolia* (Jacq.) Marechal, 1969, and *V. unguiculata,* to reduce damage by *C. punctiferalis.* When *R. communis* was monocropped, *C. punctiferalis* caused 53 % damage, but when intercropped with the above mentioned species, the damage was between 35 and 53 %. Sowing date is another cultural method for reducing damage and the presence of pests. Salihu *et al*. (2014) suggest that the correct time for planting *R. communis* crop must be related to the rainy season, which is more important than any other pest control measure in Africa, since the rains decrease the presence of certain pests.

Genetic control includes the use of cultivars resistant to insect pests, however, according to Singh *et al*. (2015), breeding *R. communis* is complicated by limited sources of pest resistance. In India, there are *R. communis* varieties that are tolerant or resistant to attack by pests of greater economic importance, such as *E. flavescens, T. ricini, S. litura, A. janata, C. punctiferalis,* and *L. trifolii* (Anjani *et al*. 2010; Anjani 2012). Resistant or tolerant plants have high oil content (between 40 and 49 %) and yields that oscillate between 540 and 1,580 kg/ha (Lavanya *et al*. 2012). It is mentioned that the cultivars having purple leaves are resistant to the attack of *L. trifolii*, while those with green leaves are susceptible (Sarma *et al*. 2006; Anjani *et al*. 2007). Sarma *et al*. (2006) mention that purple-leaf varieties have high levels of anthocyanin, which make the plant more tolerant to *L. trifolii* attack, and the epicuticular wax on their leaves reduces infestation and defoliation by *A. janata and S. litura.* There are hybrids, such as GCH4, that are resistant to the attack by *E. flavescens* due to the high wax content on the plant stems and leaves (Lakshmi *et al*. 2005). Five accessions *viz*., RG-43, RG-631, RG-1621, RG-3037 and RG-3067, among 165 core set accessions representing diversity in the entire collection maintained at ICAR-Indian Institute of Oilseeds Research, Hyderabad, India, exhibited resistance reaction against *E. flavescens*; oil content of these accessions was 46, 51, 51, 51, 52 %, respectively (Anjani *et al*. 2018). On the other hand, Severino *et al*. (2012) recommended parallel research to determine the increased potential susceptibility to pests in breeding programs to develop low-ricin, low-ricinine, and low-allergen cultivars to reduce hazardous chemical products found in *R. communis*.

Research is being carried out on the use of transgenic plants of *R. communis.* In India, two transgenic varieties of *R. communis,* Jyothi and VP1, developed by genetic engineering induce *A. janata* mortality above 88 % due to the *Bacillus thuringiensis* gene *CryAb* (Malathi *et al*. 2006).

A little explored method for monitoring and massive trapping of *R. communis* pests has been the use of pheromones, kairomonal attractants and light traps. In India, the pheromone compounds of some pests of economic importance have been identified and used for monitoring and massive trapping of *C. punctiferalis, S. litura, Amsacta albistriga* Walker, 1864 (Lepidoptera: Arctiidae), *A. janata,* and *S. obliqua* (Cork and Hall 1998). In this country, an important prerequisite for successful management of *S. litura*, the most destructive insect pest of *R. communis* damaging the crop from July- October during the south-west monsoon (kharif season), has been the implementation of an intensive monitoring program of *S. litura* population using sex pheromone traps (Satyagopal *et al*. 2014). Setting twelve traps baited with pheromone compounds per hectare for massive trapping of *S. litura* is recommended (Nandagopal and Rathod 2007; Raghavaiah 2011). In Brazil, researchers are now taking the first steps toward identifying the pheromone compounds of *C. gossypii* (Fregadolli *et al*. 2012) with the aim of developing a commercial pheromone. In India, the kairomonal compounds of the most destructive lepidopteran insect pest of *R. communis*, such as *S. litura*, *A. janata*, and *C. punctiferalis* have been identified for trapping. In field experiment, water trap baited with phenyl acetaldehyde + 2-phenyl ethanol recorded significantly higher moth catches of *S. litura* (6.8 moths/trap/wk) and *C. punctiferalis* (5.8 moths/trap/wk) (Duraimurugan *et al*. 2017). Recently, Duraimurugan and Alivelu (2018), determined the relationship of pheromone trap catches corresponding to the economic threshold level of 25 % defoliation of *S. litura* on *R. communis*, which was estimated to be 81.4 moths/trap/week. Light traps using ultraviolet black-blue spectrum have also been suggested to capture *Phyllophaga* sp. adults as a measure of ethological control Saldarriaga Cardona *et al*. (2011).

Biological control (spraying entomopathogenic microorganisms and releasing entomophagous insects) has been implemented in the control of key *R. communis* pests in countries such as India and Colombia. In India, for example, parasitism rates between 10.4 and 28.7 % of *M. maculipennis* and *Cotesia* sp. were recorded on larvae of *A. janata* and *S.*

litura, respectively (Suganthy 2007), while 9.5 % parasitism rates of *Cotesia flavipes* (Cameron, 1891) against *S. obliqua* larvae have been observed (Yadav *et al*. 2010). Also, in India, Rajasekhar *et al*. (1999) mention that when *Chrysoperla carnea* (Stephens, 1836) were released*,* the *T. urticae* (= *T. telarius*) mite populations diminished by 75 %. In the case of *S. litura*, the release of 150 adults of the parasitoid *Telenomus remus* (Nixon, 1937) per egg mass and the release of the larval parasitoid *Campoletis chlorideae* Uchida, 1957 in a parasitoid: host ratio of 1:15 achieved parasitism rates above 96 % (Satyanarayana *et al*. 2005).

Biological control through entomopathogenic nematodes, bacteria, fungi, and virus exposed to the principal pests of *R. communis* has been evaluated in India. For instance, mortality of *S. litura* pupae was evaluated with two nematode species: *Heterorhabditis indica* Poinar, Karunaka and David, 1992, and *Steinernema carpocapsae* (Weiser, 1955) (Raveendranath *et al*. 2008). Other studies evaluated the mortality of *A. janata* and *Samia ricini* (Drury, 1773) larvae exposed to two species of bacteria: *Bacillus thuringiensis* Berliner, 1915 and *Bacillus cereus* Frankland & Frankland 1887 (Manson *et al*. 1954; Kattegoudar *et al*. 1994; Mathur *et al*. 1994; Vimala Devi and Sudhakar 2006). Duraimurugan *et al*. (2015) conducted research to determine the mortality of *A. janata* larvae and *C. punctiferalis* adults with *Beauveria bassiana* (Balsamo) Vuillemin, 1912 fungus. Mortality of *A. janata* larvae exposed to granulovirus was also evaluated (Naveen Kumar *et al*. 2013). In Colombia, Saldarriaga Cardona *et al*. (2011) reported that the control of *Phyllophaga* sp. larvae was achieved by applying the *B. popilliae* Dutky bacterium at a concentration of 24,000.00 billion spores/ha a year, during five consecutive years.

The use of secondary metabolites derived from plants and other organisms, as well as methods of chemical control, have been assessed for controlling some *R. communis* pests. In laboratory studies, it has been found that methanol extracts of *Clathria longitoxa* (Hentschel, 1912) and *Callyspongia diffusa* (Ridley, 1884), two marine sponges, have insecticidal effect on *A. janata* and *P. recini* larvae (Joseph *et al*. 2010). Furthermore, *Calotropis gigantea* (L.) W. T. Aiton, 1811, leaf extracts demonstrated strong feeding deterrent activity against larvae of *Pericallia ricini* (Fabricius, 1775) at high concentrations (Neelanarayanan and Indira 2010). Likewise, toxic effects and strong antifeedant activity of raw acetonic extracts of *Mormodica charantia* L., 1753, *Tectona grandis* L. f., 1790, and *Madhuca indica* J. F. Gmel., (1791) against *S. litura* and *A. janata* larvae (Devanand and Usha Rani 2008) were found. Neem, *Azadirachta indica* (Juss, 1830), also evaluated for the control of *S. litura*, induces mortality of larvae at high concentrations (Choudhury and Aizur Rahman 2008).

Chemical control through insecticides is one of the most common practices for control of *R. communis* pests (Gahukar 2018). In Colombia, six pesticides for the control of *C. gossypii* were evaluated. The results of insect control efficacy three days after pesticide application were as follows: (from the least to the most effective) thiamethoxam + lambda-cihalotrin (0.00 %), spinetoram (0.00 %), malathion (20.35 %), thiamethoxam (38.62 %), dimethoate (86.94 %), and imidacloprid (87.33 %); whereas after seven days the following results were obtained: thiamethoxam + lambda-cihalotrin (0.00 %), spinetoram (21.46 %), malathion (38.77 %), thiamethoxam (50.84 %), dimethoate (86.14 %), and imidacloprid (90.37 %) (Varón *et al*. 2010). Mead (1989) suggested the use of carbaryl or malathion for controlling *C. gossypii* in Florida, USA. In Colombia, Saldarriaga Cardona *et al*. (2011) recommended application of baits poisoned with carbaryl at a dose of 2 to 3 g/L for the control of *A. ipsilon* and *S. frugiperda*; the same authors recommended application of liquid chlorpyrifos at the base of the plants at a dose of 1.5 - 2.0 cc/L.

The most recommendable strategy of *R. communis* pest control is Integrated Pest Management (IPM). Most of the IPM programs have been directed against key pests of *R. communis,* such as *S. litura, C. punctiferalis,* and *A. janata* (Prabhakar *et al*. 2003; Singh *et al*. 2006; Basappa 2009). In India, the growers increased seed production of *R. communis* up to 28 %, by implementing IPM programs with insecticides, crop rotation, insect traps, application of neem extract, and intercropping (Basappa 2007). The results of research in India demonstrate that IPM is an efficient strategy for the control of *A. janata* and *S. litura*, two of the key pests of *R. communis*. It is possible to decrease populations of these pests by using the recommended IPM program, which includes the use of bird perches for predatory birds to rest and to look for preys, foliar applications of 5 % neem seed extracts, biological insecticide consisting of nuclear polyhedrosis virus (*S. litura* NPV 100 LE/ha), monocrotophos at 0.5 %, and manual removal of larvae (Suganthy 2010). The pest control effectiveness of carbaryl 50W 0.2 %, endosulfan 35 EC 0.05 %, triazophos 40 EC 0.05 %, spinosad 45 SC 0.018 %, fipronil SSC 0.01 %, extract of neem seeds 5 % (weight/volume), *B. thuringiensis* 0.1 %, and a control without applying the dose of 500 L/ha, was evaluated under field conditions 30 and 45 days after establishing a plantation of a *R. communis* variety susceptible to leafminer *L. trifolii.* The results showed that the least damage (lowest number of insect mines) was found when spinosad and triazophos were applied and, at the same time, the best yield was obtained with both treatments (883 and 835 kg seed/ha, respectively) (Akashe *et al*. 2009). On the other hand, natural enemy impact has been proven to be greatest at sites adopting biointensive IPM (BIPM); par example, studies conducted by Basappa (2009) shown that BIPM modules were safer to *A. janata* eggs (*T. chilonis*) and larvae (*M. maculipennis*) parasitoids with 16.1 and 66.1 % average field parasitism, compared to chemical pesticide intensive integrated pest management modules with 6.9 and 21.2 % parasitism, respectively.

Conclusions

There is a wide range of arthropods that damage *R. communis* in different parts of the world where this plant is cultivated; many of these are considered pests of economic importance. Likewise, there are reports of a great variety of natural enemies, which have been used in biological control programs. According to the coevolutive hypothesis, it was found that the greatest richness and abundance of arthropods associated with *R. communis* is in Asia and Africa, considered as the center of origin of this plant. Most phytophagous arthropods feed on leaves. The natural enemies with more abundance and richness are the parasitoids that mostly attack the larvae of phytophagous arthropods. With respect to pollinators, *A. mellifera* is the principal pollinating insect, however, more research on pollination and floral ecology in *R. communis* is needed, in order to determine what other floral visitors may act as pollinators, and how they can be protected or manipulated to increase crop yield. The pest management programs of phytophagous arthropods of *R. communis* must be directed toward promoting and preserving natural enemies and pollinating insects by means of environment-friendly pest management techniques, for which use of wide-spectrum insecticides must be avoided.

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Author contribution

Guillermo López-Guillén, Jaime Gómez Ruiz and Juan F. Barrera defined the content of the study, conducted the literature review and wrote the manuscript. All authors read and approved the final manuscript.