



Review

Insect meal as renewable source of food for animal feeding: a review

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ABSTRACT

The massive utilisation in animal feeding of soy or fishmeal poses severe environmental issues. The insects could be a sustainable protein source. This article documents 150 species of insects that are currently commercially available in the EPP0 (European and Mediterranean Plant Protection Organization) region and in North America. Furthermore, the various data regarding body composition are analysed. Amino acids and fatty acids of several insect species are compared with the composition of soy and fishmeal as principal protein sources for animal feeding. As a protein source, insects, depending on the species, have an adequate profile of amino acids. The more frequent limiting amino acids are histidine, lysine, and tryptophan, which could be incorporated into the diet. In conclusion, insects appear to be a sustainable source of protein with an appealing quantity and quality and acceptable nutritive properties. In conclusion, the use of insects as a sustainable protein rich feed ingredient in diets is technically feasible, and opens new perspectives in animal feeding.

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1. Introduction

Feed was identified as the major contributor to land occupation, primary production use, acidification, climate change, energy use and water dependence (Mungkung et al., 2013). The productions of fishmeal or soy meal are mainly linked to these impacts.

The nutritive needs of monogastric species, particularly fish, include a high quality and quantity of protein in the diet. From a nutritional point of view, protein sources must have a high-protein content, an adequate amino acid profile, high digestibility, good palatability and no anti-nutritional factors (Barrows et al., 2008). Fishmeal and soy meal are the two of the most useful protein sources in animal feeding.

Fishmeal is obtained from the fishery. The fish catch data refer to the total global landings in 2009; 22.8 million tons of fish caught were for non-food uses (25.7%). Of this volume, 17.9 million tons (20.2% of total) were transformed into fishmeal and oil meal (FAO, 2012). Currently, the world production of fishmeal and fish oil has stabilised at approximately 5.0–6.0 million tons per year. The soybean crop is one of the most widespread in the world and grew by 1.4% from 2010 to 2011. In 2009, soybean production reached

210.9 million tons, which is slightly more than 50% of the total production of oilseed meal (FAO, 2012).

Soy meal has a high digestibility, high quality and quantity of protein and the best amino acid profile of the vegetable protein sources available, together with other nutritive benefits. Vegetable feedstuffs have several unfavourable characteristics, such as imbalances between essential and nonessential amino acids, anti-nutritional factors, low palatability and a high proportion of fibre and non-starch polysaccharides, which limit the percentage of inclusion in the diet. Under these conditions, it becomes necessary to complete the diet by adding amino acids or a high value protein source, such as animal protein, which has high digestibility and a good balance between essential and nonessential amino acids (Refstie et al., 1997, 1998; Webster et al., 1992, 1995).

Fishmeal has the advantages of animal protein sources, together with excellent nutritive properties that are indispensable, particularly for fish. These advantages, together with the current laws forbidding the use of most meat meals due to problems of food security, make fishmeal the most used animal protein source.

However, the massive utilisation of soy or fishmeal poses severe environmental issues. On one hand, increased soy cultivation causes the deforestation of areas with a high biological value (Carvalho, 1999; Osava, 1999), high water consumption (Steinfeld et al., 2006), the utilisation of pesticides and fertilisers (Carvalho, 1999), and transgenic varieties (Garcia and Altieri, 2005), which cause significant environmental deterioration (Osava, 1999). On the other hand,

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Abbreviation list

A	adult
AAE	essential aminoacids
AANE	no essential aminoacids
ARA	arachidonic acid (20:4n – 6)
CP	crude protein
DHA	docosahexaenoic acid (22:6 n3)
E	eggs
EE	ether extract
EPA	eicosapentaenoic acid (20:5 n3)
EPPO	European and Mediterranean Plant Protection Organization
FCR	feed conversion ratio

HUFA	highly unsaturated fatty acids
L	larvae
MUFA	mono unsaturated fatty acid
N	ninpha
NA	not available
ND	not detected
NFE	nitrogen-free extract
NPU	net protein utilisation
O	order
P	pupae
PER	protein efficiency ratio
PUFA	polyunsaturated fatty acids
SGR	specific growth rate

fishmeal is a resource that depends on the catch, therefore its production is quantitatively and qualitatively variable (FAO, Animal Feed Resources Information System). In addition, the deterioration of the marine environment and stripping of fisheries have resulted in a decrease in fishmeal production and an increase in the price from US\$ 600/metric ton in 2005 to US\$ 2000/metric ton in June 2010. This trend to increase prices is likely to continue (International Monetary Fund, 2010), with consequent economic repercussions on animal production. This situation reveals the importance of renewable sources of proteins, which are particularly important in the diet of fish, because fish require a high percentage of protein in their diet, either in fishmeal or through another protein source (Manzano-Agugliaro et al., 2012).

Currently, insects are being considered as a new protein source for animal feed (Premalatha et al., 2011). There are approximately one million known species of insects, although it has been estimated that their global diversity is as high as 80 million (Erwin, 2004). Grimaldi (Grimaldi and Engel, 2005) suggested that only approximately 20% of insects have been named and described. More than 58% of the known global biodiversity are insects (Footitt and Adler, 2009).

When using insects as a food source for animal feed there are several factors to consider, which include the natural feeding habits of many species, such as poultry, pig, and all cultivated species of fish, including invertebrates, such as oligochaeta, crustaceans and insects (Bell et al., 1994; Khan and Panikkar, 2009; Matsuno et al., 1999; Matthews, 1998). Insects have different feeding habits and can be fed by-products (slaughter house, restaurant surpluses, cereal remnants, etc.), whose elimination has an economic and environmental cost; insects can be reared under different conditions to optimise their nutritive value (Sealey et al., 2011). Some insect species can be grown on organic side streams, reducing environmental contamination and transforming waste into high-protein feed that can replace increasingly more expensive compound feed ingredients, such as fish meal. Then, from an environmental point of view, insect cultures are sustainable; culturing insects is usually performed in warehouses, with no need for large areas or much water, particularly when compared with crops. In addition, culturing insects contributes to the recycling of waste. On the other hand, insect by showed relative low scores on carbon footprint (Blonk et al., 2008). In addition, insects are efficient food converters because they do not use energy to maintain a high body temperature (Nijdam et al., 2012).

Due to the only recent interest in the use of insects as an alternative protein source, the nutritive properties are not well known. Previous studies on the nutritive composition of insects have focused on human nutrition, and most of those insects demonstrate a good composition for use as human food (Banjo et al., 2006; Ramos-Elorduy, 1997;

Ramos-Elorduy et al., 1982). Recent studies even mention insect development as a source of protein for human consumption for space missions (Katayama et al., 2008). Nevertheless, the utilisation of insects in animal feeding has been less studied; insects exhibit great potential for development as a standard ingredient in animal feeding. This review analyses the potential use of insect meal in animal feeding, studying the nutritive values, currently cultured species and the published findings regarding their use in animal feeding.

2. Mass-rearing of insects

The utilisation of insect meals in animal feeding requires the mass production of insects, ensuring a significant production of insects that are necessary for animal food production. Although mass-rearing has been developed for some insect species, no other insect cultures have been developed, most likely because of a lack of demand. The culture of insects is complicated because insects have strict environmental (temperature and humidity), feeding and population requirements, particularly during reproduction (Leppla, 2002). The culture of insects can be partial (from egg to larva or nymph and adult) or complete (egg–egg). The easier insects to culture are the small, multivoltine herbivores that are terrestrial, with low environmental requirements, such as pest species of crops or stocked product or gardens (Leppla, 2002). In this manner, mass-rearing has been developed, particularly for silk production, fishing bait, and pet food (Schabel, 2010).

The integrated and biological control of crop pests has provoked an interest in the knowledge of insect biology and the development of culture systems. One example is industrial warehouses that produce *Bactrocera cucurbitae* in Okinawa, Japan, which produce 40 million larvae per week (Mitsuhashi, 2010).

Cultured species include butterflies and moths (O. Lepidoptera, more than 300 species have been raised), beetles (O. Coleoptera, more than 200 species), flies and mosquitoes (O. Diptera, approximately 200 species), bugs (O. Heteroptera, less than 100 species) and bees and wasps (O. Hymenoptera, less than 100 species). Crickets and grasshoppers (O. Orthoptera), lacewings (O. Neuroptera), cockroaches (O. Blattodea), termites (O. Isoptera) and fleas (O. Siphonaptera) are also bred (approximately 10–20 species) (Leppla, 2002).

An example of mass-rearing in Table 1 indicates insects that have been used for the biological control of arthropod pests, which are or have been commercially available in the EPPO zone (European and Mediterranean Plant Protection Organization) (EPPO, 2010) and North America (Hunter, 1997) used for the biological control of arthropod pests, which are or have been commercially available in the EPPO zone (European and Mediterranean Plant Protection Organization).

Table 1
Insects which are (or had been) commercially available in the EPP0 region (EPP0, 2010) and in North America as biological control agents (Hunter, 1997).

Specie	Order	Breeding area
<i>Adalia bipunctata</i>	Coleoptera	EPP0
<i>Agrilus hyperici</i>	Coleoptera	North America
<i>Aleochara bilineata</i>	Coleoptera	EPP0
<i>Aphthona cyparissiae</i>	Coleoptera	North America
<i>Aphthona flava</i>	Coleoptera	North America
<i>Aphthona lacertosa</i>	Coleoptera	North America
<i>Aphthona nigriscutis</i>	Coleoptera	North America
<i>Apion fuscirostre</i>	Coleoptera	North America
<i>Apion ulicis</i>	Coleoptera	North America
<i>Brachyterolus pulicarius</i>	Coleoptera	North America
<i>Cassida rubiginosa</i>	Coleoptera	North America
<i>Ceutorhynchus litura</i>	Coleoptera	North America
<i>Chilocorus baileyi</i>	Coleoptera	EPP0
<i>Chilocorus bipustulatus</i>	Coleoptera	EPP0
<i>Chilocorus circumdatus</i>	Coleoptera	EPP0
<i>Chilocorus nigrita</i>	Coleoptera	EPP0
<i>Chrysolina quadrigemina</i>	Coleoptera	North America
<i>Coccinella septempunctata</i>	Coleoptera	EPP0
<i>Coleomegilla maculata</i>	Coleoptera	North America
<i>Coleomegilla maculata</i>	Coleoptera	EPP0
<i>Cryptolaemus montrouzieri</i>	Coleoptera	EPP0
<i>Cryptolaemus montrouzieri</i>	Coleoptera	North America
<i>Delphastus catalinae</i>	Coleoptera	EPP0
<i>Delphastus pusillus</i>	Coleoptera	North America
<i>Eustenopus villosus</i>	Coleoptera	North America
<i>Harmonia axyridis</i>	Coleoptera	North America
<i>Hippodamia convergens</i>	Coleoptera	North America
<i>Larinus planus</i>	Coleoptera	North America
<i>Longitarsus jacobaeae</i>	Coleoptera	North America
<i>Microlarinus lareynii</i>	Coleoptera	North America
<i>Microlarinus lypriformis</i>	Coleoptera	North America
<i>Oberea erythrocephala</i>	Coleoptera	North America
<i>Rhinocyllus conicus</i>	Coleoptera	North America
<i>Rhyzobius lophanthae</i>	Coleoptera	EPP0/North America
<i>Rhyzobius ventralis</i>	Coleoptera	North America
<i>Rodolia cardinalis</i>	Coleoptera	EPP0
<i>Scymnus rubromaculatus</i>	Coleoptera	EPP0
<i>Stethorus picipes</i>	Coleoptera	North America
<i>Stethorus punctillum</i>	Coleoptera	EPP0
<i>Stethorus punctillum</i>	Coleoptera	North America
<i>Trichosirocalus horridus</i>	Coleoptera	North America
<i>Aphidoletes aphidimyza</i>	Diptera	EPP0/North America
<i>Bangasterius orientalis</i>	Diptera	North America
<i>Cystiphora schmidti</i>	Diptera	North America
<i>Episyrrhus balteatus</i>	Diptera	EPP0
<i>Feltiella acarisuga</i>	Diptera	EPP0/North America
<i>Spurgia esulae</i>	Diptera	North America
<i>Urophora affinis</i>	Diptera	North America
<i>Urophora cardui</i>	Diptera	North America
<i>Urophora quadrifasciata</i>	Diptera	North America
<i>Urophora sirunaseva</i>	Diptera	North America
<i>Zeuxidiplosis giardi</i>	Diptera	North America
<i>Aceratoneuromyia indica</i>	Hymenoptera	North America
<i>Anagrus atomus</i>	Hymenoptera	EPP0
<i>Anagrus epos</i>	Hymenoptera	North America
<i>Anagrus fusciventris</i>	Hymenoptera	EPP0
<i>Anagrus pseudococci</i>	Hymenoptera	EPP0
<i>Anaphes iole</i>	Hymenoptera	North America
<i>Anastatus tenuipes</i>	Hymenoptera	North America
<i>Anisopteromalus calandreae</i>	Hymenoptera	North America
<i>Aphelinus abdominalis</i>	Hymenoptera	EPP0/North America
<i>Aphidius colemani</i>	Hymenoptera	EPP0/North America
<i>Aphidius ervi</i>	Hymenoptera	EPP0/North America
<i>Aphidius matricariae</i>	Hymenoptera	EPP0/North America
<i>Aphytis diaspidis</i>	Hymenoptera	EPP0
<i>Aphytis holoxanthus</i>	Hymenoptera	EPP0
<i>Aphytis lingnanensis</i>	Hymenoptera	EPP0
<i>Aphytis melinus</i>	Hymenoptera	EPP0/North America
<i>Aprostocetus hagenowii</i>	Hymenoptera	EPP0/North America
<i>Bracon hebetor</i>	Hymenoptera	EPP0/North America
<i>Bracon kirkpatricki</i>	Hymenoptera	North America
<i>Cales noacki</i>	Hymenoptera	EPP0
<i>Coccophagus lycimnia</i>	Hymenoptera	EPP0
<i>Coccophagus rusti</i>	Hymenoptera	EPP0

Table 1 (continued)

Specie	Order	Breeding area
<i>Coccophagus scutellaris</i>	Hymenoptera	EPP0
<i>Comperiella bifasciata</i>	Hymenoptera	EPP0
<i>Comperia merceti</i>	Hymenoptera	North America
<i>Cotesia flavipes</i>	Hymenoptera	North America
<i>Cotesia marginiventris</i>	Hymenoptera	EPP0
<i>Cotesia melanoscelus</i>	Hymenoptera	North America
<i>Cotesia plutellae</i>	Hymenoptera	North America
<i>Dacnusa sibirica</i>	Hymenoptera	EPP0/North America
<i>Diachasmimorpha longicaudata</i>	Hymenoptera	North America
<i>Diaeretiella rapae</i>	Hymenoptera	EPP0/North America
<i>Diglyphus isaea</i>	Hymenoptera	EPP0/North America
<i>Encarsia citrina</i>	Hymenoptera	EPP0
<i>Encarsia deserti</i>	Hymenoptera	North America
<i>Encarsia formosa</i>	Hymenoptera	EPP0/North America
<i>Encyrtus aurantii</i>	Hymenoptera	EPP0
<i>Encyrtus infelix</i>	Hymenoptera	EPP0
<i>Eretmocerus californicus</i>	Hymenoptera	North America
<i>Eretmocerus eremicus</i>	Hymenoptera	EPP0
<i>Eretmocerus mundus</i>	Hymenoptera	EPP0
<i>Goniozus legneri</i>	Hymenoptera	North America
<i>Gyranoidea litura</i>	Hymenoptera	EPP0
<i>Leptomastidea abnormis</i>	Hymenoptera	EPP0
<i>Leptomastix dactylopii</i>	Hymenoptera	EPP0/North America
<i>Leptomastix epona</i>	Hymenoptera	EPP0
<i>Lysiphlebus testaceipes</i>	Hymenoptera	North America
<i>Metaphycus flavus</i>	Hymenoptera	EPP0
<i>Metaphycus helvolus</i>	Hymenoptera	EPP0/North America
<i>Metaphycus lounsburyi</i>	Hymenoptera	EPP0
<i>Metaphycus swirskii</i>	Hymenoptera	EPP0
<i>Microterys nietneri</i>	Hymenoptera	EPP0
<i>Muscidifurax raptor</i>	Hymenoptera	North America
<i>Muscidifurax raptorellus</i>	Hymenoptera	North America
<i>Muscidifurax raptoroides</i>	Hymenoptera	North America
<i>Muscidifurax zaraptor</i>	Hymenoptera	North America
<i>Nasonia vitripennis</i>	Hymenoptera	EPP0/North America
<i>Opius pallipes</i>	Hymenoptera	EPP0
<i>Pediobius foveolatus</i>	Hymenoptera	North America
<i>Pentalitomastix plethoricus</i>	Hymenoptera	North America
<i>Praon volucre</i>	Hymenoptera	EPP0
<i>Pseudaphycus angelicus</i>	Hymenoptera	North America
<i>Pseudaphycus maculipennis</i>	Hymenoptera	EPP0
<i>Scutellista caerulea</i>	Hymenoptera	EPP0
<i>Spalangia cameroni</i>	Hymenoptera	North America
<i>Spalangia endius</i>	Hymenoptera	North America
<i>Spalangia nigroaenea</i>	Hymenoptera	North America
<i>Tetracnemoidea peregrina</i>	Hymenoptera	EPP0
<i>Tetracnemoidea pretiosa</i>	Hymenoptera	EPP0
<i>Thripobius javae</i>	Hymenoptera	EPP0
<i>Thripobius semiluteus</i>	Hymenoptera	North America
<i>Trichogramma brassicae</i>	Hymenoptera	EPP0/North America
<i>Trichogramma cacoeciae</i>	Hymenoptera	EPP0
<i>Trichogramma dendrolimi</i>	Hymenoptera	EPP0
<i>Trichogramma evanescens</i>	Hymenoptera	EPP0/North America
<i>Trichogramma exiguum</i>	Hymenoptera	North America
<i>Trichogramma minutum</i>	Hymenoptera	North America
<i>Trichogramma platneri</i>	Hymenoptera	North America
<i>Trichogramma pretiosum</i>	Hymenoptera	North America
<i>Trichogrammatoidea bactrae</i>	Hymenoptera	North America
<i>Chrysoperla carnea</i>	Neuroptera	EPP0/North America
<i>Chrysoperla comanche</i>	Neuroptera	North America
<i>Chrysoperla rufilabris</i>	Neuroptera	North America
<i>Franklinothrips megalops</i>	Thysanoptera	EPP0
<i>Franklinothrips vespiformis</i>	Thysanoptera	EPP0
<i>Karnyothrips melaleucus</i>	Thysanoptera	EPP0
<i>Scolothrips sexmaculatus</i>	Thysanoptera	North America
<i>Anthocoris nemoralis</i>	Hemiptera	EPP0
<i>Anthocoris nemorum</i>	Hemiptera	EPP0
<i>Deraeocoris brevis</i>	Hemiptera	North America
<i>Geocoris punctipes</i>	Hemiptera	North America
<i>Macrolophus caliginosus</i>	Hemiptera	North America
<i>Macrolophus pygmeus</i>	Hemiptera	EPP0
<i>Orius albidipennis</i>	Hemiptera	EPP0
<i>Orius insidiosus</i>	Hemiptera	North America
<i>Orius laevigatus</i>	Hemiptera	EPP0

Table 1 (continued)

Specie	Order	Breeding area
<i>Orius majusculus</i>	Hemiptera	EPPO
<i>Orius tristicolor</i>	Hemiptera	North America
<i>Picromerus bidens</i>	Hemiptera	EPPO
<i>Podisus maculiventris</i>	Hemiptera	EPPO/North America
<i>Xylocoris flavipes</i>	Hemiptera	North America
<i>Mantis religiosa</i>	Dipterygota	North America
<i>Tenodera aridifolia sinensis</i>	Dipterygota	North America
<i>Agonopterix alstroemeriana</i>	Lepidoptera	North America
<i>Aplocera plagiata</i>	Lepidoptera	North America
<i>Coleophora klimeschiella</i>	Lepidoptera	North America
<i>Coleophora parthenica</i>	Lepidoptera	North America
<i>Leucoptera spartifoliella</i>	Lepidoptera	North America
<i>Metzneria paucipunctella</i>	Lepidoptera	North America
<i>Tyria jacobaeae</i>	Lepidoptera	North America

3. Nutritional values

The previous studies about the nutritive value of insects show a variable proximal composition; the crude protein, lipids, nitrogen-free extract and ash vary among species or phases of development, as shown in Table 2.

3.1. Proteins

The percentage of protein is one of the most important criteria to be considered for feed protein sources. Studies regarding protein values of insects indicate that most species have a high quality and quantity of protein (Ladrón de Guevara et al., 1995; Ramos-Elorduy et al., 1981, 1982, 1984, 1997).

Table 2 displays the crude protein (CP) analysis for several species of insects. The insects are sorted in order of increasing crude protein (CP). Twenty species were identified with similar CP to that of fishmeal (CP = 60–80%), and another 28 with similar or increased (CP >40%) proportions of protein compared with that of soy meal (CP = 45–50%). The highest values were observed in the coleopteran *Metamasius spinolae* (69.1%) and *Rhantus atricolor* (71.1%), in the dipterous *Drosophila melanogaster* (70.1), as well as in *Boopedon flaviventris* (76.0%), *Melanoplus mexicanus* (77.1%) and *Sphenarium histrio* (74.8%) of the Orthoptera order.

3.2. Amino acids

Animals do not have a true protein requirement; animals actually have an amino acid requirement, which proteins are composed of (Teles et al., 2011). Therefore, one criterion to define protein quality is the amino acid composition, particularly the balance between essential and nonessential amino acids (Conconi et al., 1984). Insects have good quality amino acids are rich in essential amino acids, whereas vegetable protein sources are usually deficient lysine, methionine and leucine (Hall, 1992), which are the more frequent limiting amino acids.

As shown in Table 3, certain insect species (*Bombyx mori*, *Pachilus gigas*, *Euschistus egglestoni*, *Atizies taxcoensis*, *Musca domestica*, *Ephydra hians*) are higher in methionine than fishmeal. High levels of leucine were also detected in *B. mori*, *Cossus redenbachit*, *B. flaviventris*, *Sphenarium histrio*, *Callipogon barbatum*, *Scyphophorus acupunctatus*, *M. domestica*, *E. hians*. Nevertheless, none of the studied species have a superior level of lysine to that of fishmeal.

None of the species shown in Table 3 raise the amino acid composition of fishmeal; although *B. mori* appears to have a more adequate composition, with deficiencies only in histidine, lysine and tryptophan. However, only some species of insect have been

analysed, and we must take into account that there are millions of insect species with different stages of development that could present different amino acid profiles.

3.3. Lipids and fatty acids

Feeds rich in protein include some amount of lipids. To determine the fatty acid composition of these lipids is important because lipid contributions can meet the requirement of essential fatty acids or limit the biological function of fatty acids. On the other hand, the final composition of animal lipids reflects the fatty acid composition of the diet, which affects the quality and healthy properties of the product, such as muscle.

Depending on the ability of insects to synthesise or elongate fatty acids, four groups of insects have been described (Blomquist et al., 1991; Bondari and Sheppard, 1981; Stanley-Samuelson et al., 1988): the first one includes insects that can synthesise PUFAS de novo. These insects have a desaturase (2Δ), which allows synthesis 18:2 (n – 6), as well as elongation and desaturation until 20:3 and 20:4 (n – 6), respectively. Another group includes insects that have a lipid metabolism similar to vertebrates; these insects cannot synthesise 18:2 (n – 6) or 18:3 (n – 3), and these insects need to include these fatty acids in their diet. *Galleria mellonella* is included in this group, which cannot synthesise 18:2 (n – 6) and 18:3 (n – 3); however, *G. mellonella* can elongate and desaturate these fatty acids until 20:4 (n – 6) and 20:5 (n – 3), respectively. The final group includes mosquitoes, which require ARA and cannot elongate or synthesise di or tri-unsaturated fatty acids of 18 carbons. The fourth group, which is represented by *D. melanogaster*, apparently does not need or synthesise PUFAS.

Table 4 shows the fatty acids of different species compared with fish and soy oils. The data that are recorded in the table display the variable composition of fatty acids between species and development stages. The more common fatty acids are 18:0 and 16:0; within the monosaturated fatty acids 18:1, n9 are present in all species. Regarding PUFAS, some species of terrestrial insects (*Tenebrio molitor*, *Zophoba morio*, *B. mori*, *Acheta domesticus*, and *Locusta migratoria*) are rich in 18:2 n6 and variables of 18:3 n3 without 20:3 n6 and 20:4 n6, whereas other species are rich in 20:3 n6 and 20:4 n6, lower 18:2 n6 and 18:3 n3. The main difference between terrestrial and aquatic insects is the presence of 20:5 n3 and the absence of 20:3 n6 and 20:4 n6.

In comparison with soy oil, the meal of insects shows minor levels of 18:2 n6. However, fishmeal has high levels of HUFAS, especially 20:5 n3 (EPA) and 22:6 n3 (DHA). EPA and DHA have important biological functions in vertebrates, and it is necessary to include EPA and DHA in the diet, particularly for fish, because the velocity of synthesis is low compared with the nutritional requirements. Terrestrial insects are clearly deficient in 20:5 n3 (EPA) and 22:6 n3 (DHA), whereas 20:5 n3 is present in aquatic insects. Therefore, aquatic insects are considered a good source of this fatty acid for fresh water fish (Sushchik et al., 2003), which constitutes a large portion of the fish diet. According to Yang (Yang et al., 2006), the diet of aquatic insect larvae includes algae, which have small amounts of HUFAS, and also contain enzymes, such as ω_5 desaturase and ω_6 desaturase, which are necessary for their synthesis. These authors propose aquatic insects as the source of C20 HUFAS. However, 22:6 n3 appears only as a low percentage in a few species of the genus Notonecta and the order Ephemeroptera (Bell et al., 1994) (Table 4).

The poor quality of insect lipids can be modified, either in the manufacturing process of the meal, or during the breeding of the insect. The quality and quantity of lipids varies with developmental stages and can be modified during culture (Ghioni et al., 1996; Raksakantong et al., 2010; Stanley-Samuelson et al., 1988).

Table 2

Proximate body composition described for different insect species. CP: crude protein; EE: ether extract; and NFE: nitrogen-free extract.

Specie	Order	Phase	CP	EE	NFE	Ash	References
<i>Periplaneta americana</i>	Bl		53.9	28.4		3.3	Bernard et al. (1997)
<i>Analeptes trifasciata</i>	Co	L	20.1	2.1	70.7	1.5	Banjo et al. (2006)
<i>Aplagiognatus spinosus</i>	Co	L	25.8	36.4	19.5	3.3	Ramos-Elorduy et al. (1998)
<i>Arophalus rusticus</i>	Co	L	20.1	56.1	17.0	1.7	
<i>Callipogon barbatum</i>	Co	L	41.0	34.0			Ramos-Elorduy et al. (2006)
<i>Chalcophora</i> sp.	Co	L	30.5	53.8			
<i>Macroductylus lineaticollis</i>	Co	L	63.8	11.8			
<i>Melolontha</i> sp.	Co	L	47.4	18.8			
<i>Metamasius spinolae</i>	Co	L	69.1	17.4	9.2	0.6	Ramos-Elorduy et al. (1998)
<i>Oileus rimador</i>	Co	L	20.9	46.5			Ramos-Elorduy et al. (2006)
<i>Oryctes boas</i>	Co	L	26.0	1.5	38.5	1.5	Banjo et al. (2006)
<i>Pachymerus nucleorum</i>	Co	L	33.1	49.3			Ramos-Elorduy et al. (2006)
<i>Passalus punctiger</i>	Co	L	27.0	44.1			
<i>Paxilus leachei</i>	Co	L	21.3	47.2			
<i>Phyllophaga</i> sp.	Co	L	42.6	24.0			
<i>Rhantus atricolor</i>	Co	L	71.1	6.4	5.7	4.6	Ramos-Elorduy et al. (1998)
<i>Rhynchophorus palmarum</i>	Co	L	25.8	38.5			(Cerdeja et al., 1999)
<i>Rhynchophorus phoenicis</i>	Co	L	28.4	31.4	48.6	2.7	Banjo et al. (2006)
<i>Scyphophorus acupunctatus</i>	Co	L	35.5	51.7	5.9	1.4	Ramos-Elorduy et al. (1998)
<i>Sprataegus aloeus</i>	Co	L	47.1	17.1			Ramos-Elorduy et al. (2006)
<i>Tenebrio molitor</i>	Co	P	53.1	36.7			
	Co	P	54.6	30.8		3.4	Bernard et al. (1997)
	Co	L	47.2	43.1	0.3	3.1	Finke (2002)
	Co	L	49.5	38.1		2.8	
	Co	L	47.8	38.3			Ramos-Elorduy et al. (2006)
	Co	L	52.7	32.8		3.2	Bernard et al. (1997)
	Co	A	66.3	14.9	3.9	3.3	Finke (2002)
	Co	A	63.7	18.4		3.1	Bernard et al. (1997)
<i>Trichoderes pini</i>	Co	L	41.1	36.7	9.0	3.8	Ramos-Elorduy et al. (1998)
<i>Zophoba morio</i>	Co	L	46.8	42	2.6	2.4	Finke (2002)
<i>Copestylum anna</i>	Di	L	37.2	10	32	8.3	Ramos-Elorduy et al. (1998)
<i>Drosophila melanogaster</i>	Di	A	70.1	12.6		4.6	Bernard et al. (1997)
	Di	L	40.3	29.4		9.8	
<i>Ephydra hians</i>	Di	L	35.9	35.9	6.6	12.3	Ramos-Elorduy et al. (1998)
<i>Eristalis</i> sp.	Di	L, E, P	40.7	11.9	8.2	26.0	
<i>Musca domestica</i>	Di	L, E, P	37.5	19.8	19.6	23.1	Ogunji et al. (2008b)
	Di	L	56.7	13.5		5.0	Sheppard et al. (2007)
	Di	L	47.1	25.3		6.3	Aniebo and Owen (2010)
	Di	L	56.8	20.0		6.8	Bernard et al. (1997)
	Di	L	59.5	6.7	11.5	14.2	Djordjevic et al. (2008)
	Di	P	58.3	15.8		6.8	Bernard et al. (1997)
<i>Hermetia illucens</i>	Di	L	42.1	34.8	1.4	14.6	Newton et al. (1977)
	Di	L	37.8	31.5		13.5	Sheppard et al. (2007)
	Di	L	37.0	18.8		17.5	Ramos-Elorduy et al. (1998)
<i>Ephemera</i> sp.	Ep	N	58.7	10.5	6.1	1.9	
<i>Abedus</i> sp.	He	A	67.7	6.2	6.6	3.1	
<i>Edessa montezumae</i>	He	A, N	37.5	45.9	2.1	3.7	
<i>Euchistus strennus</i>	He	A, N	41.8	41.7	0.0	3.1	
<i>Pachilis gigas</i>	He	A, N	65.4	19.4	2.5	3.3	
<i>Apis mellifera</i>	Hy	L	41.7	18.8	34.8	3.4	
	Hy	P	49.3	20.2	24.3	3.6	
	Hy	P, L	40.5	20.3		3.4	Finke (2002)
	Hy	L, E, P	21.0	12.3	73.6	2.2	Banjo et al. (2006)
<i>Liometopum apiculatum</i>	Hy	L, P	37.3	42.1	7.8	3.1	Ramos-Elorduy et al. (1998)
	Hy	L, P	41.7	36.2	17.6	2.4	
<i>Mischocyttarus</i> sp.	Hy	L, P,	57.3	24.3	6.5	4.2	
<i>Myrmecosistus melliger</i>	Hy	A	9.5	5.8	77.7	4.1	
<i>Pogonomyrmex barbatus</i>	Hy	L, E, P	45.8	34.3	7.9	9.3	
<i>Polybia occidentalis</i>	Hy	L, P,	61.6	18.7	12.7	3.5	
<i>Polistes canadensis</i>	Hy	L, P,	61.5	31.1	1.8	1.9	
<i>Vespula</i> sp.	Hy	L, P,	52.8	29.7	11.0	3.4	
<i>Aegiale luesperiaris</i>	Le	L	40.3	29.9	22.0	3.9	
<i>Anaphe infracta</i>	Le	L	22.0	15.2	66.1	1.6	Banjo et al. (2006)
<i>Anaphe reticulata</i>	Le	L	23.0	10.2	64.6	2.5	
<i>Anaphe venata</i>	Le	L	25.7	23.2	55.6	3.2	
<i>Bombyx mori</i>	Le	L	53.8	8.1	25.4	6.4	Finke (2002)
<i>Catasticta teutila</i>	Le	L	59.8	19.2	6.7	7.1	Ramos-Elorduy et al. (1998)
<i>Cirina forda</i>	Le	L	20.2	14.2	66.6	1.5	Banjo et al. (2006)
	Le	L	33.1	12.2		7.1	Akinnawo and Ketiku (2000)
<i>Comadia redtembacheri</i>	Le	L	29.0	43.3	20.6	0.6	Ramos-Elorduy et al. (1998)
<i>Eucheira sopcialis</i>	Le	L	48.8	22.7	15.2	3.3	
<i>Heliothis zea</i>	Le	L	42.0	29.0	21.0	3.9	

Table 2 (continued)

Specie	Order	Phase	CP	EE	NFE	Ash	References
<i>Galleria mellonella</i>	Le	L	38.8	58.6		2.2	Finke (2002)
	Le	L	42.4	46.4		2.7	Bernard et al. (1997)
	Le	A	55.2	6.6		10.8	
<i>Lanifera cyclades</i>	Le	L	45.9	30.3	14.2	4.6	Ramos-Elorduy et al. (1998)
<i>Ostrinia nubilalis</i>	Le	L	60.4	17.2		2.9	Bernard et al. (1997)
	Le	P	64.2	17.0		2.6	
<i>Phasus triangularis</i>	Le	L	13.2	77.2	3.0	1.4	Ramos-Elorduy et al. (1998)
<i>Macrotermes bellicosus</i>	Is	A	20.4	28.2	43.3	2.9	Banjo et al. (2006)
<i>Aeschna multicolor</i>	Od	N	52.3	16.7	6.2	12.9	Ramos-Elorduy et al. (1998)
<i>Anax</i> sp.	Od	N	56.2	22.9	3.0	4.2	
<i>Acheta doméstica</i>	Or	A	66.6	22.1		3.6	Finke (2002)
	Or	A	64.9	13.8		5.7	Bernard et al. (1997)
	Or	N	67.2	14.4	3.9	4.8	Finke (2002)
<i>Boopedon flaviventris</i>	Or	A, N	76.0	8.4	2.3	3.0	Ramos-Elorduy et al. (1998)
<i>Melanoplus mexicanus</i>	Or	A	77.1	4.2	4.0	2.4	
<i>Schistocerca</i> sp.	Or	A, N	61.1	17.0	7.0	4.6	
<i>Sphenarium histrio</i>	Or	A, N	74.8	8.6	2.6	3.5	
<i>Sphenarium purpurascens</i>	Or	A, N	52.6	19.6	14.5	2.3	
<i>Trimerotropis pallidipennis</i>	Or	A, N	62.9	22.2	2.6	4.8	
<i>Zonocerus variegatus</i>	Or	A	26.8	3.8	63.2	1.2	

L = larvae; P = pupae; N = ninpha; E = eggs.

Bl – Blattodea, Co – Coleoptera; Di – Diptera; Ep – Ephemeroptera; He – Hemiptera; Ho – Homoptera; Hy – Hymenoptera; Is – Isoptera; Le – Lepidoptera; Me – Megaloptera; Od – Odonata; Or – Orthoptera; Pl – Plecoptera; and Tr – Trichoptera.

3.4. Digestibility

Another characteristic of protein quality is the digestibility of the source. Ramos-Elorduy et al. (1981) determined the digestibility of several of edible insect species of Mexico, and the digestibility varies among species from 77.9% to 98.9% for organic matter and from 45% to 66.9% for proteins, which composes more than 60% for most species.

Longvah et al. (2011) compared the protein digestibility of several protein sources in rats. The digestibility for eri silkworm

(*Samia ricinii*) was 86%, which is much higher than the value of 57% for canned pinto beans, 66% for canned chickpeas, 52% for peanut meal and 40% for whole wheat.

3.5. Indigestible components: chitin

Chitin is a polysaccharide present exclusively in the exoskeleton of arthropods. Chitin is a polymer of $\beta(1 \rightarrow 4)$ joined by a $\beta(1 \rightarrow 4)$ glycosidic bond, which is a crude fibre (Lindsay et al., 1984), and, in consequence, is not digestible by monogastric animals.

Table 3

Amino profile of fish meal and some insects (g/100 g dry weight). Higher values have been identified in bold.

Order	Specie	Phase	AAE							AANE							References				
			Arg	His	Ile	Leu	Lys	Met	Phe	Thr	Trp	Val	Asp	Cys	Glu	Gly		Pro	Ser	Tyr	Ala
Lepidoptera	<i>Samia ricinii</i>	P	4.4	2.7	4.4	6.6	6.5	2.3	5.2	4.8	NA	5.4	9.9	0.5	13	4.9	6.5	5.3	6.4	6.1	Longvah et al. (2011)
	<i>Bombyx mori</i>	P	6.8	2.5	5.7	8.3	6.5	4.6	5.1	5.4	0.9	5.6	11	1.4	15	4.6	4	4.7	5.4	5.5	Rao (1994)
	<i>Cossus redtenbachi</i>	L	6	1.6	5.1	7.9	4.9	2.1	9.3	4.7	0.6	6.1	11	1.3	17	5.5	5.9	6.2	5.3	6.5	Ramos-Elorduy et al. (1982)
Hemiptera	<i>Pachilis gigas</i>		4.1	7	4.2	6.9	4.5	3.6	14	3.6	0.6	6.2	6.6	2.4	10	7	7.7	4.5	5.8	8.7	Ramos-Elorduy et al. (1997)
	<i>Euschistus egglestoni</i>		4.3	3.2	4.4	7	3	2.8	3.3	4.8	0.6	6.1	7.1	1	9.6	6	6.7	3.9	4.8	9.3	Ramos-Elorduy et al. (1982)
	<i>Atizies taxcoensis</i>	NA	5.1	1.8	4.1	7.7	3.1	2.7	10	4.2	0.1	7.3	8.5	1	11	7	8.9	6.1	6.6	12	Ramos-Elorduy et al. (1982)
Orthoptera	<i>Boopedon flaviventris</i>		4.3	2.4	4.7	8.8	5.5	1.8	4.1	4.4	0.6	5.7	8.8	2	15	7.5	6.8	4.3	7.4	5.9	Ramos-Elorduy et al. (1997)
	<i>Gryllus testaceus</i>		3.7	1.9	3.1	5.5	4.8	1.9	2.9	2.8	NA	4.4	6.3	1	9.1	3.6	4.5	3.7	3.9	5.6	Wang et al. (2005)
	<i>Sphenarium histrio</i>	NA	6.6	1.1	5.3	8.7	5.7	2	12	4	0.6	5.1	9.3	1.3	4.3	5.3	7.2	5.1	7.3	7.7	Ramos-Elorduy et al. (1982)
	<i>Callipogon barbatum</i>		5.9	2.2	5.8	10	5.7	2	4.7	4	0.7	7	9.1	2	10	9.2	6.2	3.7	4.2	8	Ramos-Elorduy et al. (2006)
Coleoptera	<i>Scyphophorus acupunctatus</i>		4.4	1.5	4.8	7.8	5.5	2	4.6	4	0.8	6.2	9.1	2.2	16	6.1	5.4	6.6	6.4	6.5	Ramos-Elorduy et al. (1997)
	<i>Vespa basalis</i>		1.7	1.1	2.6	3.5	1.9	0.9	1.9	1.8	NA	2.6	3.4	ND	7.5	3.6	3.7	1.9	2.5	3.4	Ying and Long (2010)
Diptera	<i>Polistes sagittarius</i>		1.6	1.1	2	2.8	1.6	0.5	1.8	1.5	NA	2.4	3	ND	6.2	2.5	3.2	1.6	1.8	2.6	
	<i>Musca domestica</i>	L	5.2	2.9	4.4	7.8	7.3	4.6	13	4.4	0.6	5.1	11.1	2.4	13	5.8	4.8	3.7	7	6.5	Ramos-Elorduy et al. (1982)
	<i>Ephydra hians</i>	L	2.7	1	5	8	5.8	3.8	10	4.6	0.4	5.6	11	2.2	16	4.9	6.5	3.8	5.1	12	
Guide	Good quality fish meal		6.14	3.6	4.8	7.8	7.9	2.5	4.1	4.4	1	5.2	9	1	13	6.2	4.5	4	3.2	6.3	Lall and Anderson (2005)

AAE: essential aminoacids, AANE: no essential aminoacids, NA: not available, and ND: not detected.

Phase: L = larvae; P = pupae; N = ninpha; E = eggs; and A = adult.

The chitin composition varies among species and development phases. In field crickets, the percentage of chitin content is 8.7% (Wang et al., 2004), and, in larvae of *Cirina forda*, the percentage of chitin content is 9.4% (Akinawo and Ketiku, 2000; Bergeron et al., 1988; Ramos-Elorduy et al., 1997). In total, 66.6 and 11.6 mg chitin/kg dry matter in silk worms and in a brood of bees were obtained, respectively (Finke, 2007).

Chitin interferes with the use of protein (Longvah et al., 2011). The elimination of chitin in adult honey bees (*Apis mellifera*) increased the NPU (net protein utilisation) from 42 to 62, which was similar to casein, with an NPU of 70 (Ozimek et al., 1985).

Nevertheless, high digestive chitinase levels are associated with fish, such as cobia (*Rachycentron canadum*), that lack mechanical structures to break down chitinous exoskeletons of prey (Lindsay et al., 1984). Chitin, with a caloric content of 17.1 kJ g⁻¹, could constitute a substantial percentage of the total energy intake for wild cobia (Gutowska et al., 2004). Chitinase genes have been sequenced in marine teleosts, including the Japanese flounder (*Paralichthys olivaceus*) (Kurokawa et al., 2004) and the striped bass (*Morone saxatilis*) (GenBank accession no. EU048546), confirming that several marine carnivorous teleost are capable of the endogenous production of chitinase.

Additional benefits of incorporating chitin into marine fish diets have been reported, including an increase in the activity of the innate immune system in sea bream (*Sparus aurata*) (Esteban et al., 2001), the stimulation of macrophage activity in rainbow trout (*Oncorhynchus mykiss*) (Sakai et al., 1992), and increased growth rates and assimilation efficiencies in aquacultured fish (Kono et al., 1987).

Chitin may have a positive effect on the functioning of the immune system not only in fish. By feeding insects to chickens, the use of antibiotics in the poultry industry – which may lead to human infection with drug-resistant bacterial strains – may be diminished (Huis et al., 2013). A review of the immunological response to chitin and its possible role in inducing asthma and allergies revealed that the responses appeared to depend on the particle size of the chitin substance; in other words, medium-sized chitin particles induce allergic inflammation, while small-sized chitin particles may have the reverse effect of reducing the inflammatory response (Brinchmann et al., 2011). The consequences for the pathogenesis of asthma and allergies following increased consumption of chitin through the promotion of insects as food are unpredictable. However, if allergies are catalysed by a lack of exposure to chitinous substances in childhood, as suggested, increasing the consumption of insects in early childhood could, by extension, support better protection against allergies later in life (Huis et al., 2013). However chitin removal increases the quality of insect protein to a level comparable to that of products from vertebrate animals (Belluco et al., 2013).

4. Species feeding diets containing insects

4.1. Poultry

A major problem facing the poultry industry is the provision of feeds that will contain all the necessary diet components for birds to grow rapidly within a short period (Oyegoke et al., 2006). There is a need for supplementing cereal-based rations with a high quality animal protein. The importance of fishmeal as a feed ingredient in poultry production is enormous (Ijaiya and Eko, 2009). The presence of fishmeal in a complete poultry diet will supplement any amino acid deficiency in vegetable proteins, such as soybean, groundnut cake and cotton seed cake (Miles and Jacobs, 1997).

Adult, larval and pupal forms of insects are consumed naturally by wild birds and free-range poultry (Zuidhof et al., 2003). The insects that are used for fowl feeding have a long history; however,

the scale is small, between 2% and 15%. Before the 1990s, in the eastern Sichuan area, duck breeders always collected the housefly larva *M. domestica* for duck feed. Mass-rearing insect species are primarily the flour weevil *T. molitor* L., black powder insect *Tenebrio obscurus*, barley insect *Zophobas morio* and the dipteran housefly larva, which are widely used for feeding chickens, ducks and some other animals (Yi et al., 2010).

The species of the order Orthoptera are naturally consumed by wild birds and free-range poultry. However, Nakagaki and DeFoliart (1987) noted that the inclusion of methionine and arginine improve the FCR (feed conversion ratio) in poultry feed with *A. domesticus*. In general, feeding experiments with various species of Orthoptera have shown positive results, without adding amino acids. Thereby, the inclusion of 15% grasshopper (*Acridia cinerea*) meal in broiler chicken feed does not affect growth (Wang et al., 2007). This result coincided with reports by other researchers, which did not find differences in growth indexes between broilers that were fed the control diet, a cricket-based diet, *Anabrus simplex* (Finke et al., 1985) or *Gryllus testaceus* (Wang et al., 2005), or with grasshopper meal (Ojewola et al., 2005). Similar results were obtained by Finke et al. (1985), even with high levels (28%) of substitution.

Lepidoptera meal has also been used in broiler feed, Oyegoke et al. (2006) replacing fishmeal (4% of the diet) with larvae of *C. forda*, and determined that there were no significant differences between the growth performance of broiler chicks that were fed the compounded larval diets and that of broiler chicks that were fed the conventional fishmeal. Meanwhile, Ijaiya and Eko (2009), using *Anaphe infracta* (7%) observed that there were no significant differences in performance, in terms of the feed intake, body weight gain, feed conversion efficiency and protein efficiency ratio, across the dietary treatments (with fishmeal or silkworm meal).

Within O. Diptera, most of the experimental diets were based on *M. domestica*. Pro (Pro et al., 1999) determined that the protein and energy that was supplied by dry fly larvae (*M. domestica*) support animal performance similar to that of chicks that were fed a conventional diet of sorghum-soybean. Neither Awoniyi et al. (2003), Djordjevic et al. (2008) nor Ocio et al. (1979) found significant differences in weight increase between birds that were fed diets containing *M. domestica* larvae or high quality fishmeal. Têguia et al. (2002) also determined that the total weight gain in the control group that exclusively received fishmeal was significantly lower than that of birds that received the diet containing the largest amount of maggot meal. All these studies suggest that compounded insect diets contain a protein source that is comparable in quality to that present in the conventional fishmeal that is used for broiler feed (Oyegoke et al., 2006).

However, Ijaiya and Eko (2009) demonstrated that intake was reduced because of the inability of young chicks to effectively utilise the crude fibre inherent in the exoskeleton (made of chitin) of the silkworm caterpillar. This finding agrees with the report of Fagoonee (1983), who suggested that the complete substitution of SCM for FM in broiler chicks reduced consumption due to the high oil and fibre content of silkworm caterpillars.

Concerning the quality of the meat, Ojewola et al. (2005) found that fishmeal exhibited no superiority over any of the other diets (with grasshopper meal) in terms of the carcass quality of broiler chickens that were produced. Hwangbo et al. (2009) found that chicken feeding diets containing 10–15% maggots (substituting soy) can improve the carcass quality and growth performance of broiler chickens. In addition, there was no adverse effect on the taste of the meat from birds that were fed a corn-cricket (*A. simplex*) diet, with 28% insect meal, as determined by a taste panel (Finke et al., 1985).

Economic value is another factor to consider when employing insects as food. Ijaiya and Eko (2009) determined that the total cost

Table 4
Relevant Fatty acids (% of total fatty acids) of terrestrial and aquatic insects.

	Order	Stage	14:00	16:00	18:00	SFA	16:01	18:1n9	MUFA	18:2 n6	18:3 n3	20:3 n6	20:4n6	20:5n3	22:6n3	PUFA	References
Fish oil			3.2	13.5	2.7		9.8	23.7		1.4	0.6		1.6	11.2	12.6		NRC (1993)
Soy oil			0.1	10.3	3.8		0.2	22.8		51	6.8						
Terrestrial insects																	
<i>Copris nevinsoni</i>	Co	L	0.3	1.31	28.26	31.72	ND	3.68	3.68	1.75	0	10.36	40.24	12.94	ND	65.27	Raksakantong et al. (2010)
<i>Holotrichia</i> sp.	Co	A	ND	0.77	27.92	29.42	0.51	5.59	6.11	ND	0	13.86	47.26	ND	3.39	64.5	
<i>Tenebrio molitor</i>	Co	L	3.3	16.1	2.5	22.3	3	41.9	44.9	30.9	1.4	0	0	0	0	32.3	Finke (2002)
<i>Tenebrio mollitor</i>	Co		1.7	18.6	5.7	26.9	1.3	39.1	40.4	29.9	1.1	0	0	0	0	31	
<i>Zophoba morio</i>	Co		1	31	7.4	40.2	0.4	38.8	39.2	19.3	0.6	0	0	0	0	19.9	
<i>Meimuna opalifera</i>	Ho		2	2.47	52.53	56.98	0.28	0.92	1.2	ND	0	10.77	33.03	ND	ND	43.8	Raksakantong et al. (2010)
<i>Tessarotoma papillosa</i>	He	ND	0.46	41	41.46	ND	7.27	7.27	ND	0	4.67	46.68	ND	ND	53.35		
<i>Oecophylla smaragdina</i>	Hy		0.4	1.68	31.41	34.59	ND	1.96	1.96	ND	0	5.82	57.73	ND	ND	63.55	
<i>Termes</i> sp.	Hy		0.1	0.78	31.9	32.83	0.19	1.86	2.06	0.34	0	8.9	56.01	ND	ND	65.25	
<i>Macrotermes bellicosus</i>	Is	A	2.17	42.5	2.86	47.53	2.1	15.84	17.94	24.24	3.9	0	4.94	0	0	33.08	Ekpo and Onigbinde (2007)
<i>Bombyx mori</i>	Le	L	2	13.8	9.7	29.6	0.8	25.9	26.7	28.3	11.3	0	0	0	0	39.6	Finke (2002)
<i>Bombyx mori</i>	Le	P	0.2	20.9	7.4	28.8	0.7	31.2	31.9	19.1	18.5	0	0	0	0	37.6	Katayama et al. (2008)
<i>Cirina forda</i>	Le	L	0.7	13	16	31.14	0.2	13.9	14.1	8.1	45.3	0.04	0.04	0.04	0.04	53.56	Akinawo and Ketiku (2000)
<i>Galleria mellonella</i>	Le	L	0.2	34.6	1.5	36.45	2.2	54	56.2	6.6	0.5	0	0	0	0	7.1	Finke (2002)
<i>Acheta doméstica</i>	Or	A	0.6	24.7	9.2	35.6	1.4	24.4	25.8	36.3	1	0	0	0	0	37.3	
<i>Acheta doméstica</i>	Or	N	0.7	21.6	10.3	34.6	1.1	22.7	23.8	39	1.4	0	0	0	0	40.4	
<i>Brac. portentosus</i>	Or		ND	1.61	35.79	37.54	0.71	3.4	4.11	ND	ND	7.94	50.43	ND	ND	58.37	Raksakantong et al. (2010)
<i>Locusta migratoria</i>	Or	A	2.3	39	5.4	46.7	1.8	26.2	28	3.8	21.4	0	0	0	0	25.2	Beenackers and Scheres (1971)
Aquatic insects																	
<i>Berosus</i> sp.	Co	L+A	3.1	18.1	15.4	37.7	6.7	19.7	26.4	11.3	4.9	0	0	5.7	0	21.9	Hanson et al. (1985)
<i>Lara avara</i>	Co	L+A	0.9	18.5	2.1	21.8	9.1	36.2	45.3	21.3	5.5	0	0	0.9	0	27.7	
<i>Psephenus</i> sp.	Co	L+A	3	21.7	3	29.4	12	24.6	36.6	5.4	7	0	0	9.7	0	22.1	
<i>Tropisternus</i> sp.	Co	L+A	3.6	17	9.4	31.4	7.5	18.1	25.6	10.3	10.9	0	0	8.4	0	29.6	
<i>Beetle</i> sp.	Co	L	1.3	17.1	7.2	25.6	10.4	12.1	22.5	6.1	11.8	0	1.7	15.6	0.01	35.21	Bell et al. (1994)
<i>Agathon</i> sp.	Di	L	2.2	20.6	13.5	36.5	5.1	17.7	22.8	4	16.6	0	0	7.1	0	27.7	Hanson et al. (1985)
<i>Atherix variegata</i>	Di	L	6.2	13.7	4.8	24.9	12	21.3	33.3	6.1	10.4	0	0	15.3	0	31.8	
<i>Chironomidae</i> sp.	Di		2.3	17.9	5.1	25.3	21.9	9.1	31	6.6	5.1	0	1.1	11.6	0.1	24.5	Bell et al. (1994)
<i>Amoletus</i> sp.	Ep	L	2.3	22.5	2	27	25.2	19.1	44.3	2.5	5.2	0	0	14.9	0	22.6	Hanson et al. (1985)
<i>Attenella delantaia</i>	Ep	L	2.1	20.8	6.8	30	17.7	16.3	34	4	10.4	0	0	13.6	0	28	
<i>Baetis</i> sp.	Ep	L	1.7	23.5	5.1	30.4	9.8	14.8	24.6	4.3	6.6	0	0	24.7	0	35.6	
<i>Gen Caenis</i>	Ep	N	2.6	22.1	2.7	27.4	23.1	7.4	30.5	2	3.8	0	0.9	15.6	0	22.3	
<i>Cinygmula</i> sp.	Ep	L	1.1	11	5.9	18.2	16.5	27.6	44.1	7.7	7.3	0	0	8.4	0	23.4	
<i>Drun. coloradensis</i>	Ep	L	1.8	16.8	5	24.2	14.6	23	37.6	5.4	8.4	0	0	17.4	0	31.2	
<i>Drunella grandis</i>	Ep	L	2.3	17.6	5.1	25.9	12	19.6	31.6	3.8	11.2	0	0	20.6	0	35.6	
<i>Ecdyonur. venosus</i>	Ep	N	3	22.8	2	27.8	23.8	5.3	29.1	1.8	4.8	0	0.7	14.8	0	22.1	Bell et al. (1994)
<i>Gen Ephemerella</i>	Ep	N	3.6	22.7	2.6	28.9	19.4	7	26.4	2.4	5.3	0	0.9	15	0.02	23.62	
<i>Heptagenia</i> sp.	Ep	L	2.1	30.2	4.4	37.3	20.6	12.8	33.4	2.9	6.3	0	0	13.3	0	22.5	Hanson et al. (1985)
<i>Ironodes</i> sp.	Ep	L	3	19.4	3.1	26	23.9	17.4	41.3	4.6	3.4	0	0	14.4	0	22.4	
<i>Isonydia</i> sp.	Ep	L	1.5	27.5	3.8	33.1	11.5	19.4	30.9	4	11.2	0	0	12.6	0	27.8	
<i>Paraleptophe.</i> sp.	Ep	L	3.5	21.5	3.5	28.8	20.1	27.4	47.5	6.3	3.6	0	0	5.5	0	15.4	
<i>Rhithrogena</i> sp.	Ep	L	1.3	19.4	2.8	23.5	25.9	18.3	44.2	6.1	10.3	0	0	5.9	0	22.3	
<i>Tricorythodes</i> sp.	Ep	L	1.8	22.6	5.9	30.5	15.7	20.8	36.5	3.2	3.3	0	0	12.3	0	18.8	
<i>Gen Notonecta</i>	He		1.8	15.3	6.2	23.3	13.7	10.6	24.3	6.1	11.3	0.01	0.7	12.7	0.1	30.91	Bell et al. (1994)
<i>Corixidae</i> sp	He		0.6	15.8	5.7	22.1	4.7	9.1	13.8	9.4	27.2	0.2	2	9.9	0.7	49.4	
<i>Sialis</i> sp.	Me	L	1.4	10.3	6.9	20.2	7.6	22	29.6	13.9	5	0	0	14.2	0	33.1	Hanson et al. (1985)
<i>Anomala hastatum</i>	Od	L	6.5	25.5	18.8	50.8	4.4	17	21.4	7.9	3.3	0	0	2.6	0	13.8	
<i>Argia</i> sp	Od	L	2.2	18.5	6.5	27.8	12	25.1	37.1	5	7	0	0	18	0	30	
<i>Hesperope pacifica</i>	Od	L	1.6	13.1	5	20.7	8	23.8	31.8	6.7	11.5	0	0	19.9	0	38.1	
<i>Hesperope pacifica</i>	Od	A	1.7	14.2	3.6	20.6	9.9	28.7	38.6	4	15.8	0	0	15.3	0	35.1	
<i>Ishmura</i> sp.	Od	L	1.9	19.6	10.4	32.2	5.5	18.1	23.6	10.3	9.5	0	0	9.8	0	29.6	
<i>Isogenoides</i> sp.	Od	L	1.6	14.1	6.8	23	12.3	22.6	34.9	5.6	11	0	0	16.6	0	33.2	
<i>Isoperia</i> sp.	Od	L	1	15.6	3.6	20.6	8.8	30.1	38.9	3.5	16.6	0	0	14.6	0	34.7	
<i>Octogomphus</i> sp.	Od	L	1.6	16.4	6.4	24.8	13.9	25.5	39.4	10.1	6.3	0	0	7.4	0	23.8	

(continued on next page)

Table 4 (continued)

Order	Stage	14:00	16:00	18:00	SFA	16:01	18:1n9	MUFA	18:2 n6	18:3 n3	20:3 n6	20:4n6	20:5n3	22:6n3	PUFA	References
Pteronarcella	L	3.6	12.1	4	20.3	2.5	26	28.5	17.6	15.9	0	0	7.3	0	40.8	
Yoraperla sp.	L	1.5	18.4	4.6	25.7	3.4	30.3	33.7	15.9	10.1	0	0	6	0	32	
Stonefly sp.	PI	1.7	16.6	4.2	23.5	14.7	13.6	28.3	4.1	6.7	0	0	15.2	0.3	27.6	Bell et al. (1994)
Allocosmoecus	Pr	2.9	19	2.8	25.2	18	15.6	33.6	5.4	12.2	0	0	8.2	0	25.8	Hanson et al. (1985)
Clistoro magnifica	L	0.6	1.9	28.1	8.2	21.8	30	29.5	4.5	0	0	0.4	0	34.4		
Clistoro magnifica	Pr	0.6	25.2	2	27.9	7.6	19.9	27.5	32.1	4.9	0	0	0.5	0	37.5	
Discosmo. gilvipes	L	0.8	15.5	25.3	5.4	22	27.4	13.1	15.5	0	0	11.3	0	0	39.9	
Discosmo. gilvipes	Pr	2.2	22.3	27.4	16.8	19.5	36.3	4.7	16.9	0	0	4	0	0	25.6	
Eclisomyia sp.	L	0.6	21.5	24	4	20.7	24.7	27	11.2	0	0	0.9	0	0	39.1	
Glossosma sp.	L	3.4	23.1	29.9	15.9	12.4	28.3	5.5	9.2	0	0	14.4	0	0	29.1	
Glossosma sp.	Pr	3.7	25.4	34.6	12.8	13.8	26.6	4.5	17.7	0	0	4.8	0	0	27	
Hesperophylax sp.	L	6.8	22	31.5	14.9	14.1	29	6.8	17.8	0	0	7.9	0	0	32.5	
Heteroptec californicum	L	1.1	21.5	26.2	3.7	21.2	24.9	17.8	22.8	0	0	1.3	0	0	41.8	
Hydatophylax sp.	L	1	13	20.7	2.3	25.9	28.2	13.7	24.1	0	0	3.7	0	0	41.5	
Hydropsyche sp.	L	12	17.2	33.2	9.4	20.6	30	5.4	12.6	0	0	9.2	0	0	27.2	
Lepidostoma sp.	L	1.6	24.6	2.8	29.4	7.8	21.3	29.1	11.2	16.8	0	0	2.7	0	30.7	
Namanyia sp.	L	1.1	15.1	2.6	18.8	4.9	44.2	49.1	20.8	7.1	0	0	1.1	0	29	
Neophylax sp.	Pr	2.7	23.3	1.5	27.9	19.9	18.6	38.5	2.8	15.8	0	0	6	0	24.6	
Oncosmoecus sp.	L	0.8	14.2	2.9	18.1	2.4	15.9	18.3	35.8	11.1	0	0	2.6	0	49.5	
Parapsyche sp.	L	7.5	21	3.7	32.2	12.1	18.8	30.9	6.5	6.3	0	0	13.2	0	26	
Pseudostenophylax sp.	L	2.5	6.4	5.7	15.3	1	25.6	26.6	9.9	26.8	0	0	2	0	38.7	
Psychoglypha subborealis	L	2.3	14.5	4.9	23.5	24.7	14.7	39.4	6.3	6.7	0	0	4.3	0	17.3	
Ryacophilla sp.	L	2.9	20.1	3.4	27	13.7	21.5	35.2	7.8	6.3	0	0	14.2	0	28.3	
Ryacophilla sp.	Pr	0.9	18.1	5.9	25.7	9.1	23.4	32.5	16.9	9.5	0	0	9.6	0	36	

PUFA: polyunsaturated fatty acids; MUFA: mono unsaturated fatty acid, and ND: not detected.

Order: Co – Coleoptera; Di – Diptera; Ep – Ephemeroptera; He – Hemiptera; Ho – Homoptera; Hy – Hymenoptera; Is – Isoptera; Le – Lepidoptera; Me – Megaloptera; Od – Odonata; Or – Orthoptera; Pl – Plecoptera; and Tr – Trichoptera.

of intake as well as the total cost per kg gain gradually declined with increasing inclusion levels of silkworm caterpillar meal. This finding is in agreement with observations of other researchers (Awoniyi et al., 2003; Khatun et al., 2005, 2003; Têguia et al., 2002).

4.2. Pigs

There are few scientific studies on the use of insects as alternative feed for pigs. Only Newton et al. (1977) evaluated the feeding value and palatability of dried soldier fly larvae (*Hermetia illucens*) fed to pigs. *Hermetia* meal composed 33% of the diet and replaced soy. Although the authors did not determine production rates, the authors determined that the apparent digestibility of dry matter for pigs that were fed the larvae meal diet was significantly lower than for that of pigs that were fed the soybean meal diet. Additionally, when given a choice between diets, pigs did not discriminate against a diet containing larvae meal.

4.3. Fish

Fish are cultured animals that have higher exigencies regarding the quality and quantity of protein, with fishmeal as the essential ingredient and major protein source in fish feed. The utilisation of insects as a protein source for fish feed has been scarcely studied. Nevertheless, the interest in this topic is increasing.

In the fish *Clarias gariepinus*, the replacement of 25% of fishmeal by *Zonocerus variegatus* L. meal produces an improved growth rate and nutrient utilisation compared with fish that were fed the control diet (fishmeal based) (Alegbeleye et al., 2012). Alegbeleye et al. (2012) also obtained a superior performance compared with other diets that have higher inclusion levels (50, 75 and 100%) of *Z. variegatus* L. in terms of the final mean body weight, FCR (Feed Conversion Ratio), SGR (Specific Growth Rate) and PER (Protein Efficiency Ratio). However, growth was negatively influenced in *C. gariepinus* fingerlings when the inclusion of *Z. variegatus* L. was increased above 50%, with a significant decrease in nutritive indices. At 100% inclusion, the growth performance declined significantly (Alegbeleye et al., 2012). According to Spreen et al. (1984), low levels of chitin in the diet could improve growth efficiency through enhancement in bi-dobacterium. Nevertheless, at higher inclusion levels of *Z. variegatus* L., there was a trend towards the reduced digestibility of proteins and lipids.

Similar results were obtained in African catfish (*C. gariepinus*) that were fed slices of mealworms (*T. molitor*); a 20% substitution improved the growth and nutritive index of catfish. However, a reduction in growth performance, as well as in feed and protein utilisation, were observed in catfish that were fed high levels of mealworm meal or solely mealworms (Ng et al., 2001). Nandeesh et al. (1988) observed similar results in catla-rohu hybrids (*Catla catla* × *Labeo rohita*) and *Cyprinus carpio* that were fed diets with a 30% replacement using silkworm pupae meal.

The larvae of *H. illucens* (L.) have also been evaluated as feed in various fish species, including channel catfish and tilapia (*Oreochromis* sp.), alone and in combination (50% larvae, 50% commercial diet) with high-(45%) and low-(30%) protein commercial diets. No differences in the body weight and total length of the two species of fish were found among experimental diets (Bondari and Sheppard, 1981). In rainbow trout *O. mykiss* that were fed diets containing *H. illucens* pre-pupae, it was determined that the replacement of 25% of the fishmeal and 38% of the fish oil components of a commercial diet had no effect on the feed conversion ratio (St-Hilaire et al., 2007). The effects of replacing 25 and 50% of the fishmeal with normal *H. illucens* or fish offal-enriched *H. illucens* pre-pupae has been studied in the rainbow trout *Oncorhynchus mykiss*. The growth of fish that were fed enriched *H. illucens* diets was not

significantly different from those fish that were fed the fishmeal-based control diet, whereas the growth of fish that were fed normal *H. illucens* diets was significantly reduced compared with the control diet (Sealey et al., 2011).

The quality of the fillet is primarily affected by the fatty acid profile, which depends of the content of fish oil in the diet. The diet with black soldier fly pre-pupae reduced fish oil by 38% (i.e., from 13 to 8%); however, fish that were fed black soldier fly diets that were low in fish oil had reduced levels of omega-3 fatty acids in their muscle fillets (St-Hilaire et al., 2007). Nevertheless, no significant difference was observed in a blind comparison of fish that were fed the fishmeal-containing control diet compared with fish that were fed normal *H. illucens* or fish offal-enriched *H. illucens* pre-pupae diets (Sealey et al., 2011). Instead, in channel catfish and tilapia that were fed *H. illucens* alone, and in combination (50% larvae 50% commercial diet) with high-(45%) and low-(30%) protein commercial diets, taste was not affected, indicating that fish that are fed larvae are acceptable to the consumer (Bondari and Sheppard, 1981).

The results that were obtained in different fish species that were fed different species of insects demonstrate the potential of these protein sources for fish feed (Achionye-Nzeh and Ngwudo, 2003; Ajani et al., 2004; Aniebo et al., 2011; Fasakin et al., 2003; Kroeckel et al., 2012; Ogunji et al., 2008a; Rangacharyulu et al., 2003; Wuertz et al., 2012). Currently, the chitin content, digestibility, amino acid balance, and fatty acid composition, appear to prevent the inclusion of insects in fish diets at levels higher than 20–30%. Different strategies to enhance the nutritive values of insect meal could allow increased percentages of fishmeal substitution.

5. Conclusions

Thus far, the use of insects in animal feed has not received much attention. Recently, some studies, which have generally been developed in underdeveloped countries with a traditional use of insects as food, have been published, which have drawn the attention of the international community and have shown the nutritive potential of insects. These studies, together the development of mass-rearing systems for insects, the current economic crisis and the increase in food prices, provides interesting perspectives for the use of insects for different purposes, such as animal nutrition, agriculture, to obtain essential oils or biodiesel. Additionally, insect culturing does not compete with food resources or land use and maximises the benefits of waste management by using “waste nutrients” for insect growth. Additionally, insect utilisation contributes to the natural recycling of nutrients. Therefore, we believe that the coming years will see a significant increase in scientific production related to the use of insect meal in animal feed or other purposes. To make use of insects as a feed ingredient on a large scale it is important to increase the scale of insect production further with a continuous quantity and quality, but it should be decrease the cost price of insect rearing further in order to be competitive with currently used protein sources.

The published results regarding the use of insect meal in animal feeding indicate that insects have a great potential in animal feeding. As a protein source, insects have an adequate profile of amino acids, depending on the insect species. The more frequent limiting amino acids are histidine, lysine and tryptophan, which could be incorporated into the diet. In addition, it is necessary to evaluate the amino acid profiles of other insect species to select the species with the best amino acid profile or to improve the profile through genetic methods. To introduce insects as a feed ingredient in the feed chain, additional research is recommended on its feeding value, inclusion levels in diets, and functional properties of

the feed ingredient. In conclusion, the use of insects as a sustainable protein rich feed ingredient in diets is technically feasible, and opens new perspectives in animal feeding.

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