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Recent insights into the extraction, characterization, and bioactivities of chitin and chitosan from insects

Kannan Mohan a,b,*, Abirami Ramu Ganesan b,*,1, Thirunavukkarasu Muralisankar c, Rajarajeshwaran Jayakumar d, Palanivel Sathishkumar e, Venkatachalam Uthayakumar a, Ramachandran Chandrasekar a, Nagarajan Revathi a

a PG and Research Department of Zoology, Sri Vasavi College, Erode, Tamil Nadu, 638 316, India
b School of Applied Sciences, College of Engineering, Science and Technology (CEST), Fiji National University, 5529, Fiji
c Aquatic Ecology Laboratory, Department of Zoology, School of Life Sciences, Bharathiar University, Coimbatore, Tamil Nadu, 641 046, India
d Department of Molecular Medicine, Faculty of Medicine, University of Malaya, Kuala Lumpur, 50603, Malaysia
e Key Laboratory of Theoretical Chemistry of Environment, Ministry of Education, School of Chemistry, South China Normal University, Guangzhou, 510006, PR China

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ABSTRACT

Background: Insects are a living resource used for human nutrition, medicine, and industry. Several potential sources of proteins, peptides, and biopolymers, such as silk, chitin, and chitosan are utilized in industry and for biotechnology applications. Chitosan is an amino-polysaccharide derivative of chitin that consists of linear amino polysaccharides with \textit{D}-glucosamine and N-acetyl-\textit{D}-glucosamine units. Currently, the chief commercial sources of chitin and chitosan are crustacean shells that accumulate as a major waste product from the marine food industry. Existing chitin resources have some natural challenges, including insufficient supplies, seasonal availability, and environmental pollution. As an alternative, insects could be utilized as unconventional but feasible sources of chitin and chitosan.

Scope and approach: This review focuses on the recent sources of insect chitin and chitosan, particularly from the Lepidoptera, Coleoptera, Orthoptera, Hymenoptera, Diptera, Hemiptera, Dictyoptera, and Odonata orders. In addition, the extraction methods and physicochemical characteristics are discussed. Insect chitin and chitosan have numerous biological activities and could be used for food, biomedical, and industrial applications.

Key findings and conclusions: Recently, the invasive and harmful effects of insect species causing severe damage in agricultural crops has led to great economic losses globally. These dangerous species serve as potential sources of chitin and are underutilized worldwide. The conclusion of the present study provides better insight into the conversion of insect waste-derived chitin into value-added products as an alternative chitin source to address food security related challenges.

1. Introduction

Insects have been considered a valuable food source since ancient times, with ~2 billion people globally consuming 1900 different species of insects for human nourishment (Van Huis, 2013). Major insect consumers are in Southeast Asia, the Pacific, sub-Saharan Africa, and Latin America. In general, insects consist of 30–45% protein, 25–40% fat, and 10–15% chitin (Spranghers et al., 2017). Chitin is the second most abundant bioactive polysaccharide in nature following cellulose. Among the various components in insects, chitin is a significant biopolymer, and the extraction of chitin and chitosan from insects is more advantageous in terms of extraction methods, chemical consumption, time and yield compared to existing sources. However, the proportion of chitin varies in every species in relation to its life-cycle. Adult \textit{Tenebrio molitor} and \textit{Hermithia illincis} species contain up to 5% chitin (Marino-Pérez, 2015), whereas the prepupa/pupa stages of black soldier flies, Tebo worms, Turkestan cockroaches, and house flies contain 21 g/kg, 11.1 g/kg, 6.7 g/kg and 11.9 g/kg of chitin, respectively, which represents 1.2%

* Corresponding author.
** Corresponding author.
E-mail addresses: kmohanphd@gmail.com (K. Mohan), abirami.rg@gmail.com (A.R. Ganesan).
1 Both authors contributed equally to this manuscript.

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Chitin is considered to be a fibre with defensive activity against microbes. While the chemical chitinase is found in human gastric juices, it has been found to be inactive. Chitin is therefore, is mostly hydrolysed by lysozymes and hydrochloric acid found in human saliva and the stomach (Adamkova et al., 2017).

Recently, scientists have extracted chitin from cicada quagmires, silkworms, and honeybees and described the functional properties of chitosan from these sources (Ma, Xin, & Tan, 2015). They reported that chitosan from insect sources is promptly accessible because of their reproductive rate and their ease of cultivation. Similarly, the removal of chitosan from the original organism influences its biological activity, and the extraction of chitosan from insects can be practised utilizing moderate conditions instead of the rigid conditions required for extraction from marine crustaceans. The yield of chitosan material from insects is higher than from shellfish, and chitin and chitosan from insect species have been reported to have useful applications (Y. Zhao, Park, & Muzzarelli, 2010).

For example, chitosan extracted from cicada slough, silkworm chrysalises, mealworms, and grasshopper species showed higher potential water holding capacity (594–795%) and fat binding capacity (275–645%) compared to shrimp shell chitosan. This property is a promising feature for food applications. Additionally, C. molossus L. consists of 33 g/100 g of chitin that demonstrates better mechanical properties, including tensile strength (62 mPa) and elongation at break (10.4%), for the production of a biodegradable film similar to that of commercial medical grade shrimp chitosan film (Ma et al., 2015). Further, chitin isolated from Pterophylla beltrani showed better anti-fungal activity against the entomopathogenic fungi M. anisoplia (Torres-Castillo et al., 2015).

These studies show the benefits of using insect-based chitin/chitosan in biomedical and food applications that have recently been reported. However, conventional ethnobiological information demonstrates that insects have been used as nourishment and as an indispensable ingredient for treatments of various diseases since ancient times. Insects as traditional medicine are frequently not revealed or reported to the world (Adiletta et al., 2015).

Fig. 1. The research distribution diagram of chitin and chitosan from insects.
Hermetia illucens larvae that was defatted with CHCl₃·CH₃OH (7:3 mixture, at 20 °C for 4 h) yielded 93 g of chitin-containing material (Khayrova, Lopatin, & Varlamov, 2019), whereas demineralization (using 2% HCl at 20 °C for 2 h) and deproteinization (5% NaOH at 50 °C for 2 h) yielded 58% and 46% of chitin, respectively. Although it was reported as a maximum yield, the biotechnology industries require a single step process and green technology for the removal of fat. For example, concentrated mineral acids are used to maximize the chitin yield in a single step process. Mineral acids such as phosphoric acid do not hydrolyse the chitin, unlike HCl and H₂SO₄. This process replaces multiple-step processes such as delipidation, demineralization, or deproteinization in chitin extraction.

2.2. Deproteinization (DP)

The deproteinization step is quite difficult due to the cleavage of the chemical bonds between the chitin and proteins. Chemical treatments are the first step in the removal of proteins. Generally, a wide range of chemicals have been used for the deproteinization of commercial chitin from shrimp, crab, lobster, and krill, and reaction conditions vary considerably between studies. The chemical extraction of chitin from insects is explained in Table 1. Furthermore, NaOH is the preferential inorganic base, and it is applied in various concentrations, ranging from 0.125 to 5.0 M (Kaya et al., 2014; Kaya, Erdogan, Mol, & Baran, 2015; M. W.; Kim, Song, Han, et al., 2017; Luo et al., 2019; Soon, Tee, Tan, Rosnita, & Khalina, 2018); at varying temperatures, up to ≥160 °C (Ibitoye et al., 2018; Kaya, Lelesiú, et al., 2015; Kaya et al., 2016; Shin, Kim, & Shin, 2019; S.; Wu, 2011; Xia, Chen, & Wu, 2013); and at various treatment durations (from a few minutes up to a few days) (Luo et al., 2019; N. H. Marei, Abd El-Samie, Salah, Saad, & Elwahy, 2016; Mehranian, Pourabad, Bashir, & Taieban, 2017; Sajomsang & Gonil, 2010; Julliana Isabelle; Simionato, Villalobos, Bulla, Coró, & García, 2014; Y.
Table 1

Extraction methods, characterization and biological activities of chitin and chitosan from insects.

| Order/species                           | Deproteinization | Demineralization | Decoloration | Deacetylation | Yield (%) | Characterization | Physical properties/ Biological activities | References |
|-----------------------------------------|------------------|------------------|--------------|---------------|------------|-----------------|--------------------------------------------|------------|
| **Lepidoptera**                         |                  |                  |              |               |            |                 |                                            |            |
| Silk worm, Bombyx mori                  | 1 M NaOH in 90 °C for 2 h | 1 M HCl in 30 °C for 2 h | 2% KMnO₄ for 2 h | 60% NaOH in 100 °C for 8 h | NA | 3.1 | XRD, FT-IR, TGA, SEM | Rheological | Luo et al. (2019) |
| Bombyx mori                             | NaOH (1.0 mol L⁻¹) for 24 h at 80 °C | HC1 (1.0 mol L⁻¹) for 20 min at 100 °C | 0.4% Na₂CO₃ for 2 h | 40 % wt NaOH and NaBH₄ for 6 h | NA | NA | FT-IR, ¹³C NMR, DTG, SEM | Textile effluents treatment | Simionato et al. (2014) |
| Bombyx mori                             | NaOH (1.0 mol L⁻¹) for 24 h at 80 °C | HC1 (1.0 mol L⁻¹) for 20 min at 100 °C | 0.4% Na₂CO₃ for 2 h | 40 % wt NaOH and NaBH₄ for 6 h | 2.59 | 88.40 | FT-IR, ¹³C NMR, TGA, DTG, SEM | Textile wastewater treatment | Simionato et al. (2006) |
| Bombyx mori                             | NaOH (1.0 mol L⁻¹) for 24 h at 80 °C | HC1 (1.0 mol L⁻¹) for 20 min at 100 °C | 0.4% Na₂CO₃ for 2 h | NaOH (40 wt %), with NaBH₄ (0.83 g L⁻¹) for 8 h | 2.59 | 88.40 | FT-IR, ¹³C NMR, TGA, DTG, SEM | Textile wastewater treatment | Simionato et al. (2006) |
| Flour moth, Ephestia kuehniella         | 1 M NaOH at 85 °C for 60 min | 1 M HCl at 100 °C for 20 min | 1% KMnO₄ for 60 min | 55% NaOH at 100 °C for 6 h | NA | 9.5-10.5 | FT-IR, EA, EDX, SEM | NA | Mehrian et al. (2017) |
| Pine caterpillar, Dendrolimus punctatus  | 5% NaOH at 7 °C for 10 h | 3% HCl at 35 °C for 20 h | 11% H₂O₂ at 85 °C for 2.5 h | 55% NaOH at 100 °C for 6 h | NA | NA | FT-IR, EA, EDX, SEM | NA | Weixing (2008) |
| Butterfly, Argynnis pandora             | 2 M NaOH solution at 50 °C for 24 h | 2 M HCl at 50 °C for 24 h | Distilled water, methanol, and chloroform (4:2:1) for 10 min | 55% NaOH (w/w), 120 °C, and 4 h | NA | 31.37 | FT-IR | NA | Wu (2011) |
| Hawk moth, Clanis bilineata             | Flavourzyme hydrolysis at pH 6.5 and 50 °C | NA | NA | NA | NA | NA | FT-IR | Anti-oxidant Anti-ageing | Wu et al. (2013) |
| Clania bilineata                        | 10% (w/v) NaOH at 60 °C for 24 h | 7% (v/v) HCl at 25 °C for 24 h | NA | NA | NA | NA | FT-IR | Anti-oxidant Anti-bacterial | Wu (2011) |
| Clania bilineata                        | 10% (w/v) NaOH at 60 °C for 24 h | 7% (v/v) HCl at 25 °C for 24 h | NA | NA | NA | NA | FT-IR, TGA, XRD, SEM | NA | Kaya et al. (2015a) |
| Clania bilineata                        | 10% (w/v) NaOH at 60 °C for 24 h | 7% (v/v) HCl at 25 °C for 24 h | NA | NA | NA | NA | HPLC, FT-IR | Anti-oxidant Anti-bacterial | Kaya et al. (2015a) |
| Clania bilineata                        | 10% (w/v) NaOH at 60 °C for 24 h | 7% (v/v) HCl at 25 °C for 24 h | NA | NA | NA | NA | HPLC | Hypolipidemic | Xia et al. (2013) |
| **Coleoptera**                          |                  |                  |              |               |            |                 |                                            |            |
| Mealworm, Tenebrio molitor              | 1 M NaOH in 90 °C for 2 h | 1 M HCl in 30 °C for 2 h | 2% KMnO₄ for 2 h | 60% NaOH in 100 °C for 8 h | NA | 2.5 | XRD, FT-IR, TGA, SEM | Rheological | Luo et al. (2019) |
| Tenebrio molitor                        | 500 mL 5% NaOH at 95 °C for 3 h | 3 h in 1500 mL 2 N HCl at 20 °C | NA | 500 mL of NaOH at 95 or 105 °C for 3 h or 5 h | Dry-17.32 | 2.5 | XRD, FT-IR, TGA, SEM | Rheological | Song et al. (2018) |
| Comb-clawed beetles, Omophus sp.        | 2 M NaOH for 20 h at 100 °C | 2 M HCl for 4 h at 50 °C | Methanol-chloroform-water (21:4:1) | 50 mL of 4 M HCl solution at 75 °C for 2 h | NA | NA | SEM, XRD, TGA, FTIR | BSA adsorption capacities | Kaya et al. (2016a) |
| White grub cockchafer, Melolontha melolontha | 4 M NaOH at 150 °C for 18 h | 50 mL of 4 M HCl solution at 75 °C for 2 h | Water, alcohol and chloroform (4:21:1) for 20 min | 2 M HCl at 60 °C for 20 h | NA | NA | SEM, XRD, TGA, ESEM, EA | BSA adsorption capacities | Kaya et al. (2014b) |
| Melolontha sp.                          | 1 M NaOH for 20 h at 100 °C | 100 mL of 1 M HCl at 90 °C for 1 h | Chloroform, methanol, and water (1:2:4) | 100 mL of 2 M HCl at 65-75 °C for 2 h | NA | 13-14 | FT-IR, TGA, XRD, ESEM, EA | BSA adsorption capacities | Kaya et al. (2014b) |
| Water scavenger beetles, Hydrophilus piceus | 100 mL of 1 M NaOH at 110 °C for 18 h | 100 mL of 1 M HCl at 90 °C for 1 h | Chloroform, methanol, and water (1:2:4) | 130 mL M HCl at 80 °C for 30 min | NA | 19-20 | FT-IR, TGA, XRD, SEM | BSA adsorption capacities | Kaya et al. (2016b) |
| Colorado potato beetle, Leptinotarsa decemlineata | 50 mL of 2 M NaOH at 80-90 °C for 16 h | 100 mL of 2 M HCl at 65-75 °C for 2 h | Chloroform, methanol and water (in a ratio of 1:2:4) for 1 h | 8 M NaOH at room temperature for 24 h | Adult-20 Larvae-7 | 74 | FT-IR, TGA, XRD, SEM | Antimicrobial | Kaya et al. (2014c) |
| Dung beetle, Cathartius molossus         | 4.0 M NaOH at 90 °C for 6 h | 1 M HCl at 90 °C for 1 h | Chloroform, methanol, and water (1:2:4) | 50% NaOH (w/v) 1:20 at 100 °C for 3 h | Adult- 72 Larvae-67 | 24 | FT-IR, TGA, XRD, SEM | Rheological | Ma et al. (2015) |

(continued on next page)
Table 1 (continued)

| Order/species                        | Deproteinization | Demineralization | Decoloration | Deacetylation | Yield (%) | Characterization | Physical properties/ Biological activities | References |
|--------------------------------------|------------------|------------------|--------------|---------------|------------|------------------|------------------------------------------|-------------|
|                                      |                  |                  |              |               |            |                  |                                          |             |
| Large ground beetle, *Calosoma rugosa* | 1.0 M NaOH at 100 °C for 8 h |                  |              |               |            |                  |                                          |             |
| *Calosoma rugosa*                    | 1.0 N NaOH       | 36.5% HCl        | NA           |               |            |                  |                                          |             |
| Dark black chafer beetle, *Holorichia parallela* | 1 M NaOH         | 1 M HCl for 30 min | 1% KMnO4    | NA            |            |                  |                                          |             |
| Mealworm Beetle, *Zophobas morio* and Rhinoceros Beetle, *Allomyrina dichotoma* | NaOH at 80 °C for 24 h | 7% (v/v) HCl at 25 °C for 24 h | NA          |               |            |                  |                                          |             |
| *Zophobas morio*                     | 0.5 M, 1.0 M and 2.0 M NaOH in °C for 20 h | 1.0 M of HCl in 35 °C | Glacial acetone for 30 min | 50 wt % NaOH in °C for 30 h | 0.5 M-5.43 | 1.0 M-5.22 | 20.5 | FT-IR, SEM, TGA, DSC, XRD | Anti-oxidant | Soon et al. (2018) |
| Dung beetle                          | 2. 0-2. 5 mol•L⁻¹ NaOH, 90-100 °C, for 4-5 h | 0.8 mol L⁻¹ HCl at 70 °C | NA | 10.0-11. 25 mol L⁻¹ NaOH for 3 h | 28.7 | Na | Na | NA | WA et al. (2018) |
| *Lucanus cervus*                     | 1 M NaOH in 90 °C for 1 h | 1 M HCl in 90 °C for 1 h | chloroform-methanol-water (1:2-4, v:v) | NA | 10.9 | NA | XRD, FT-IR, TGA, SEM | NA | Kabalak et al. (2020) |
| Orthoptera                           |                  |                  |              |               |            |                  |                                          |             |
| Grasshopper                          | 1 M NaOH in 90 °C for 2 h | 1 M HCl in 30 °C for 2 h | 2% KMnO₄ for 2 h | 60% NaOH in 100 °C for 8 h | NA | 5.7 | XRD, FT-IR, TGA, SEM | Rheological | Luo et al. (2019) |
| Mexican katydil, *Poryphyla belsmani* | NA | NA | NA | NA | 11.8 | 58.8 | NA | Anti-oxidant | Torres-Castillo et al. (2015) |
| Moroccan locust, *Dociostaurus marcus* | 2 M NaOH in 50 °C for 18 h | 2 M HCl in 55 °C for 1 h | Methanol, chloroform and distilled water (in the ratio of 2:1) | 60% NaOH in 150 °C for 4 h | Nymphs-12 | Nymphs-73.8 | Adults-81.69 | FT-IR, TGA, XRD, ESEM | NA | Erdogan and Kaya (2016) |
| House cricket, *Brachytrupes portentosus* | 1 M NaOH at 95 °C for 6 h | 1 M NaOH for 20 h at 150 °C | Oxalic acid for 3 h at room temperature | 1% sodium hypochlorite for 3 h | 4.3-7.1 | Na | FT-IR, XRD, SEM | NA | Ibitoye et al. (2018) |
| *Celis variabilis*                   |                  |                  |              |               |            |                  |                                          |             |
| Decictrix verrucovorus, *Melanogryllus desertus Paracoptera labiata* |                  |                  |              |               |            |                  |                                          |             |
| *Calliptamus barbarus*               | 1 M NaOH at 80-90 °C for 21 h | 1 M HCl at 100 °C for 30 min | Chloroform:methanol:distilled water solution (1:2-4) for 1 h | 50% NaOH (w/v) at 130 °C for 2 h | 20.5 | 16.5 | 7 | FTIR, TGA, XRD, SEM | Anti-microbial | Anti-oxidant | Kaya et al. (2015b) |
| *Oedaleus decorus*                   |                  |                  |              |               |            |                  |                                          |             |
| *Allopus similarius*                 | 2 M NaOH at 175 °C for 18 h | 4 M HCl at 75 °C for 1 h | Chloroform:methanol:distilled water in the ratio of 1:2:4 | 50% NaOH (w/v) at 115 C for 2 h | 20.5 | 16.5 | 7 | ESEM, FT-IR,TGA, XRD | Anti-microbial | Anti-oxidant | Kaya et al. (2015c) |
| *Duroniella fracta*                  |                  |                  |              |               |            |                  |                                          |             |
| *Duroniella laticornis*              |                  |                  |              |               |            |                  |                                          |             |
| *Oedipoda minista*                  |                  |                  |              |               |            |                  |                                          |             |
| *Oedipoda caerulescens*              |                  |                  |              |               |            |                  |                                          |             |
| *Pygophyra cognata*                 | 1.25 M NaOH       | 2 N HCl          | NA           | 50% NaOH (w/v) | A-20.91 | B-21.68 | C-21.35 | D-23.35 | NA | NA | NA | Kim et al., 2017a |
| *Two-spotted field crickets, Gryllus bimaculatus* |                  |                  |              |               |            |                  |                                          |             |
| *Desert locust, Schistocerca gregaria* | 1.0 M NaOH at 100 °C for 8 h | 1 M HCl       | NA           | 50% NaOH (15 mL/g) at 100 °C for 8 h | 12.2 | NA | FT-IR, XRD, SEM | NA | Marei et al. (2016) |
| *Schistocerca gregaria*              | 1 M NaOH         | 1 N HCl          | NA           | 50% NaOH       | 22.5 | 25   | FT-IR, XRD | Wound healing | Marei et al. (2016) |
| *Two-spotted field crickets, Gryllus bimaculatus* | 1 M HCl in 90 °C for 1 h |                  |              |               |            |                  |                                          |             |
| Order/species | Deproteinization | Demineralization | Decoloration | Deacetylation | Yield (%) | Characterization | Physical properties/ Biological activities | References |
|---------------|------------------|------------------|--------------|--------------|------------|-----------------|------------------------------------------|------------|
| *Bradytus sureyi*  
*Gryllotalpa gryllotalpa* | 1 M NaOH in 90 °C for 14 h | Chloroform-methanol-water (1:2:4, v:v) | KMnO₄ with concentration of 1, 0.5, and 0.1% were used at 20 °C | NA | 9.8  
10.1 | NA | NA | ³¹H NMR, FT-IR | Kabalak et al. (2020) |
| **Hymenoptera** | | | | | | | | | |
| European honey bee,  
*Apis mellifera* | 1 M NaOH at 80 °C for 8 h | 2 M HCl at 80 °C for 6 h | Distilled water (40 mL), methanol (20 mL) and chloroform (20 mL) | NA | 9.8  
10.1 | NA | NA | ³¹H NMR, FT-IR | Draczynski (2008) |
| *Apsis mellifera* | 1 M NaOH at 80 °C for 8 h | 2 M HCl at 80 °C for 6 h | Distilled water (40 mL), methanol (20 mL) and chloroform (20 mL) | NA | 9.8  
10.1 | NA | NA | ³¹H NMR, FT-IR | Kaya et al. (2015d) |
| *Apsis mellifera* | 1 M NaOH at 80 °C for 8 h | 2 M HCl at 80 °C for 6 h | Distilled water (40 mL), methanol (20 mL) and chloroform (20 mL) | NA | 9.8  
10.1 | NA | NA | ³¹H NMR, FT-IR | Kaya et al. (2015d) |
| *Apsis mellifera* | 1 M NaOH at 80 °C for 8 h | 2 M HCl at 80 °C for 6 h | Distilled water (40 mL), methanol (20 mL) and chloroform (20 mL) | NA | 9.8  
10.1 | NA | NA | ³¹H NMR, FT-IR | Kaya et al. (2015d) |
| *Vespa velutina* | 1 M NaOH (100 mL) at 60 °C for 8 h | 1 M HCl (100 mL) at 50 °C for 6 h | Distilled water (40 mL), methanol (20 mL) and chloroform (10 mL) at room temperature | NA | 9.8  
10.1 | NA | NA | ³¹H NMR, FT-IR | Feis (2020) |
| Bumblebee,  
*Bombus terrestris* | 1 M NaOH at 85 °C for 24 h | 1 M HCl at 100 °C for 20 min | H₂O₂/33% HCl 9:1 | NA | 9.8  
10.1 | NA | NA | ³¹H NMR, FT-IR | Majšin et al. (2007) |
| **Diptera** | | | | | | | | | |
| Housefly,  
*Musca domestica* | 1 mol/L NaOH solution at 100 °C for 3 h  
500 mL of 1.25 N NaOH at 90 °C for 3 h | 3 h in 500 mL of 2 N HCl solution at room temperature | NA | NaOH (50% w/v) at 125 °C for 4 h  
50% NaOH at 105 °C for 3 h | 8.02  
5.87 | NA | NA | ³¹H NMR, FT-IR, XRD, DSC | Zhang et al., 2011a  
Kim et al. (2016) |
| *Musca domestica* | 1 mol/L NaOH solution at 100 °C for 3 h  
500 mL of 1.25 N NaOH at 90 °C for 3 h | 3 h in 500 mL of 2 N HCl solution at room temperature | NA | NaOH (50% w/v) at 125 °C for 4 h  
50% NaOH at 105 °C for 3 h | 8.02  
5.87 | NA | NA | ³¹H NMR, FT-IR, XRD, DSC | Zhang et al., 2011a  
Kim et al. (2016) |
| *Musca domestica* | 1 mol/L NaOH solution at 100 °C for 3 h  
500 mL of 1.25 N NaOH at 90 °C for 3 h | 3 h in 500 mL of 2 N HCl solution at room temperature | 10 mg/mL KMnO₄ for 4 h | NaOH (50% w/v) at 125 °C for 4 h  
50% NaOH at 105 °C for 3 h | 8.02  
5.87 | NA | NA | ³¹H NMR, FT-IR, XRD, DSC | Zhang et al., 2011a  
Kim et al. (2016) |
| Black soldier fly,  
*Hermetia illucens* | 1 M NaOH at 80 °C for 24 h | 1 M HCl for 1 h | 1% KMnO₄ | 400 mg/mL NaOH at 70 °C for 8 h | NA | NA | NA | Anti-oxidant  
Anti-tumour | Ai et al. (2008) |
| *Hermetia illucens* | 1 M NaOH at 80 °C for 24 h | 1 M HCl solution (250 mL) at 100 °C for 30 min | 1% potassium permanganate solution (100 IL) for 1 h | NA | 9  
23 | NA | NA | ³¹H NMR, FT-IR, XRD, TGA, SEM | Wang et al. (2020) |
| *Hermetia illucens* | 1 M NaOH 1 h at 80 °C | NA | NA | NA | 8.5  
NA | NA | NA | ³¹H NMR, FT-IR | D’Hondt et al. (2020)  
Wang et al. (2013) |
| *Hermetia illucens* | 2 M NaOH at 50 °C for 18 h | 2 M HCl at 55 °C for 1 h | NaClO at 80 °C for 4 h | NA | 3.6  
3.1  
14.1  
2.9 | NA | NA | ³¹H NMR, FT-IR, XRD, SEM | Wang et al. (2013) |

(continued on next page)
Table 1 (continued)

| Order/species | Deproteinization | Demineralization | Decoloration | Deacetylation | Yield (%) | Characterization | Physical properties/Biological activities | References |
|---------------|------------------|------------------|--------------|---------------|-----------|------------------|-------------------------------------------|------------|
| *Hermetia illucens* | NaOH at 90 °C for 3 h | HCl at 2 h | NA | NA | 21.3 | Chitin | NA | NA | ANTONOV ET AL. (2019) |
| *Hermetia illucens* | NaOH at 50 °C for 2 h | 2% HCl for 2 h at 20 °C | NA | NaOH at 100 °C for 2 h | 7 | Chitosan | 32 | NMR, FT-IR | NA | KHAYROVA ET AL. (2019) |
| *Hermetia illucens* | NA | 2 N HCl for 24 h at 15 min | NA | 9 | NA | NA | NA | NA | NA | CALIGIANI ET AL. (2018) |
| *Musca domestica* | 5% NaOH at 95 °C for 6 h | NaOH 50 °C for 2 h at room temperature for 3 h | 0.3% KMnO₄ at room temperature for 4 h | NA | 27 | Chitin | 32 | NMR, FT-IR | NA | JING ET AL. (2007) |
| *Drosophila melanogaster* | 5% NaOH at 95 °C for 6 h | NaOH 50 °C for 2 h at room temperature for 3 h | 0.3% KMnO₄ at room temperature for 4 h | NA | 27 | Chitosan | 32 | NMR, FT-IR | NA | KHAYROVA ET AL. (2019) |
| *Blowfly* | NaOH at 90 °C for 3 h | HCl at 2 h | NA | NA | 21.3 | Chitin | NA | NA | ANTONOV ET AL. (2019) |
| *Blowfly* | NaOH at 50 °C for 2 h | 2% HCl for 2 h at 20 °C | NA | NaOH at 100 °C for 2 h | 7 | Chitosan | 32 | NMR, FT-IR | NA | KHAYROVA ET AL. (2019) |
| *Blowfly* | NA | 2 N HCl for 24 h at 15 min | NA | 9 | NA | NA | NA | NA | NA | CALIGIANI ET AL. (2018) |

**Hemiptera**

| Cicada slough | 1 M NaOH in 90 °C for 2 h | 1 M HCl in 30 °C for 2 h | 2% KMnO₄ for 2 h | 60% NaOH at 100 °C for 8 h | NA | Chitin | 28.2 | XRD, FT-IR, TGA, SEM | Rheological | LUO ET AL. (2019) |
| Aquatic bug | 100 mL of 1 M NaOH at 110 °C for 18 h | 100 mL of 1 M HCl at 90 °C for 1 h | Chloroform, methanol, and water (1:2:4) | NA | 15–16 | Chitosan | 70 | FT-IR, TGA, XRD, SEM | NA | KAYA ET AL. (2014a) |
| Cicada lodosi | 2 M NaOH solution at 100 °C for 20 h | 2 M HCl for 2 h at 100 °C | Water, methanol, and chloroform mixed at the ratio of 4:2:1 | NA | 4.97 | Chitin | 6.49 | FT-IR, SEM | NA | MOL ET AL. (2018) |
| Cicada mordoganensis | 1 M NaOH in 80 °C for 36 h | 1 M HCl at 100 °C for 20 min | 6% sodium hypochlorite | NA | 36.6 | Chitin | 28.2 | XRD, FT-IR, TGA, SEM | Rheological | LUO ET AL. (2019) |
| Cicada | 1000 mL of 10% (w/w) NaOH at 60 °C for 24 h | 1000 mL of 7% (w/w) HCl at room temperature (~25 °C) for 24 h | NA | NA | 50% NaOH at 100 °C for 4 h | Chitin | 62.42 | FT-IR | Anti-bacterial | WU ET AL. (2013) |

**Dictyoptera**

| American cockroach, Periplaneta americana | 1.25 N NaOH at 95 °C for 3 h | 2 M HCl at room temperature for 3 h | CHLOROFORM, METHANOL, AND WATER | 50% NaOH at 95 °C for 3 h | 3.36 | Chitin | 2.08 | FT-IR | Anti-bacterial | KIM ET AL. (2017b) |
| Periplaneta americana | 4 M NaOH solution for 20 h at 150 °C | 4 M HCl solution for 2 h at 75 °C | WATER, METHANOL AND CHLOROFORM (IN THE RATIO OF 4:2:1) FOR 4 H AT 30 °C | NA | 2.08 | Chitin | 12.04 | FT-IR | Anti-bacterial | KAYA ET AL. (2015b) |
| Blaberus giganteus | 2 M NaOH at 90 °C for 9 h | NA | CHLOROFORM METHANOL WATER | NA | 2.08 | Chitin | 12.04 | FT-IR | Anti-bacterial | KAYA ET AL. (2017) |

(continued on next page)
### Table 1 (continued)

| Order/species       | Physical properties/activities | Degradation treatment | Characterization | References |
|---------------------|--------------------------------|-----------------------|------------------|------------|
| Odontodactylus      | Dragoney, sympetrum           | 1 M NaOH at 50°C for 1 h | NA               | Kaya et al., 2014a |
|                     |                                | 1 M HCl at room temperature for 1 h | NA               | Kaya et al., 2014b |
| Sympetrum fonscolombii| Chitin                            | 15 h                  | FT-IR, SEM       | Kaya et al., 2014a |
|                     |                                | 100 mL of 1 M NaOH in 100 mL of 1 M HCl at 110°C for 1.5 h | NA               | Kaya et al., 2014a |
| Anax imperator     | Chitin                          | 1 h                   | FT-IR, TGA, XRD  | Kaya et al., 2014a |
|                     |                                | 100 mL of 1 M NaOH in 100 mL of 1 M HCl at 110°C for 1.5 h | FT-IR, TGA, XRD  | Kaya et al., 2014a |

2.3. Demineralization (DM)

The removal of minerals, mainly using calcium carbonate, is termed demineralization. In 1978, the process of commercial demineralization of chitin from crustacean shells was patented. This process is commonly achieved by acid treatment using sulphuric, hydrochloric, nitric, acetic, oxalic and formic acids (Al Sagheer, Al-Sughayer, Muslim, & Elsabee, 2009; Srinivasan, Kanayairam, & Ravichandran, 2018). In chitin extraction from insects, HCl has been found to be superior to all of these other acids (Ibitoye et al., 2018; Mehranian et al., 2017; Percot, Viton, & Domard, 2003; Shin et al., 2019; Julliana Isabelle; Simionato et al., 2014; Y. S.; Song et al., 2018). The demineralization process involves the breakdown of calcium carbonate into calcium chloride along with the release of carbon dioxide. An alternative method to this harsh chemical demineralization is the use of lactic acid fermentation. Jung et al. (2005) demonstrated the efficacy of lactic acid fermentation for the DM of crab shell waste with *Lactobacillus paracasei* KCTC-3074 compared with chemical treatments, such as 2 N HCl, 0.1 M EDTA, and 0%–10% lactic acid.

2.4. Decolourization (DC)

The decolourization step is usually essential for removing pigments and for obtaining a colourless product. These treatments are applied to chitin sources, regardless of the nature of the starting material. The residual protein and pigments are removed for further utilization, especially for biomedical or food applications (Rinaudo, 2006). Various decolouring agents have been used for decolourization of the chitin extracted from crustacean shells and insects.

2.5. Deacetylation (DA)

Deacetylation refers to the process of eliminating the acetyl groups attached to chitin and the substitution of reactive amino groups. The degree of deacetylation determines the percent of free amino groups within the structure and would therefore be helpful in distinguishing between chitin and chitosan. DDA is taken into consideration for chitosan as it influences the physicochemical and biological properties (Nessa et al., 2010), including the acid-base ratio, electrostatic characteristics, biodegradability, self-aggregation, sorption properties, and the ability to chelate metal ions (Hussain, Iman, & Maji, 2013). Chitin can be converted into chitosan using chemical methods (Philibert, Lee, & Fabien, 2017) at industrial scale due to the feasibility of mass production. For crustacean shell waste and insects, the chemical method of deacetylation uses alkali-NaOH (Anand, Kalavani, Maruthupandy, Kumaraguru, & Suresh, 2014; N. H.; Marei et al., 2016; Paulino, Simionato, Garcia, & Nozaki, 2006; Y. S.; Song et al., 2018; Srinivasan et al., 2018; Torres-Castillo et al., 2015) or acids to deacetylate chitin. Since glycosidic bonds are highly vulnerable to acid, alkali is proposed to be a better chemical option (Haji et al., 2014). Several factors during the deacetylation reaction can impact the characteristics of the resulting chitosan product. Temperature and processing time were the parameters that had the most significant impact on the DDA and molecular weight (Rege & Block, 1999).
The disadvantages of using the traditional chitin chemical extraction process include alterations in physicochemical properties, the use of expensive chemicals in the purification process and the release of toxic effluent wastewater into the environment. These challenges lead to the deterioration of environmental health (Dhillon, Kaur, Brar, & Verma, 2013) reduce the levels of valuable proteins that can be used as animal feed (Shirai et al., 2001). Therefore, green extraction methods (Fig. 2b) are gaining popularity due to their cleaner and more eco-friendly approaches (De Holanda & Netto, 2006).

The biological extraction process using microorganisms such as Lactobacillus (Rao, Munoz, & Stevens, 2000), Pseudomonas aeruginosa K-187 (Oh, Shih, Tzeng, & Wang, 2000) and Bacillus subtilis (Yang, Shih, Tzeng, & Wang, 2000) can be used to reduced chitin degradation and reduce impurities down to a satisfactory level for specific applications. For example, Khanafari & Sanatei (2008) examined chitin and chitosan isolated from shrimp waste by chemical and microbial methods, and the results showed that the microbial process was preferable to the chemical method. The microbial method required less time, a simple procedure, and could reduce impurities down to a satisfactory level for specific applications.
low solvent consumption, and lower energy input. Although there is less research on the biological method of chitin extraction, it can replace the chemical methods that are overwhelmed with several disadvantages at the industrial scale. Other extraction methods have also been reported for chitin production, mainly from shrimp waste, including enzymatic (Gartner, Pelaye, & López, 2010), microwave-assisted (Hongkusup, Khutoryanskiy, & Niranjan, 2016) and ultrasonic-assisted (Valdez-Peña et al., 2010) and phytoextraction (Gopal et al., 2019).

Among all techniques, ionic liquids (ILs) are considered a promising volatile organic solvent for chitin production (Qin, Lu, Sun, & Rogers, 2010), although some specific ILs have some disadvantages, such as high cost and toxicity, which make them unsuitable for biological applications (Sharma, Mukesh, Mondal, & Prasad, 2013). Therefore, deep eutectic solvents (DES) are a green alternative to conventional methods of chitin production (Paiva et al., 2014). In comparison to traditional methods, DES possess more advantages, such as low or non-toxicity, lower cost, ease of synthesis and biodegradability (Q. Zhang, Vigier, Royer, & Jerome, 2012). DES extraction has been used for chitin production from shrimp (Huang, Zhao, Guo, Xue, & Mao, 2018) and lobster (Hong, Yuan, Yang, Zhu, & Lian, 2018; Zhu, Gu, Hong, & Lian, 2017), as well as in the insect Hermetia illucens (Zhou et al., 2019). Recently, Brigode et al. (2020) reported the production of chitin from H. illucens using acid detergent fibre and acid detergent lignin methods (ADF-ADL). Additional research is required to study green methods with smaller carbon-footprints for chitin and chitosan extraction from insects (Brigode et al., 2020).

### 3. Physico-chemical characterization

#### 3.1. Extraction yield

Yield is one of the crucial features in the extraction of chitin and chitosan from insects. As stated in the earlier section, the insect chitin sources have a significant amount of protein content. Therefore, deproteinization using alkaline treatments like NaOH and KOH was carried out to recover high purity chitin. The efficiency of deproteinization process depends on various factors including temperature, concentration of NaOH, and reaction time (Kaya et al., 2014; kaya, Erdogan, et al., 2015; Paulino et al., 2006). Use of high concentration of NaOH, and reaction time (Kaya et al., 2014; Kaya, Erdogan, & Sarkar, 2018), as well as in the insect Hermetia illucens (Zhou et al., 2019). Recently, Brigode et al. (2020) reported the production of chitin from H. illucens using acid detergent fibre and acid detergent lignin methods (ADF-ADL). Additional research is required to study green methods with smaller carbon-footprints for chitin and chitosan extraction from insects (Brigode et al., 2020).

### Table 3

Elemental analysis (EA) results of the insect chitin.

| Species            | Chitin (%) | References                  |
|--------------------|------------|-----------------------------|
|                    | Carbon (C) | Hydrogen (H) | Nitrogen (N) | CN ratio |
| **Bradypterus**    |            |                |             |          |
| (C.) surayai       | 46.6       | 7.7            | 5.3         | 8.8      | Kabalak et al. (2020) |
| **Gryllotalpa**    |            |                |             |          |
| gryllotalpa        | 44.2       | 7.6            | 5.0         | 8.8      |                      |
| **Polyphylla**     |            |                |             |          |
| julio             | 45.4       | 7.5            | 5.1         | 8.9      |                      |
| **Lucanus**        |            |                |             |          |
| cervus             | 45.9       | 7.6            | 5.3         | 8.5      |                      |
| **Melolontha**     |            |                |             |          |
| melolontha         | 45.09      | 6.29           | 6.72        | NA       |                      |
| **Holotrichia**    |            |                |             |          |
| parallela          | 44.36      | 5.92           | 6.45        | 6.88     |                      |
| **Cicada sloughs** |            |                |             |          |
|                   | 40.85      | 6.12           | 5.92        | NA       |                      |
| **Bumblebee**      |            |                |             |          |
|                   | 43.92      | 6.43           | 5.92        | NA       | Majtán et al. (2007) |
| **Periplaneta**    |            |                |             |          |
| americana          | 45.74      | 6.59           | 6.69        | NA       | Kaya et al. (2015b)  |
| **Hermetia**       |            |                |             |          |
| illucens           | 39.74      | 5.46           | 6.00        | 6.62     | Purkayastha and Sarkar (2020) |
| **Hermetia**       |            |                |             |          |
| illicens           | 43.74      | 5.82           | 6.14        | 7.12     |                      |
| **Vespa crabro**   |            |                |             |          |
|                   | 46.62      | 6.42           | 6.85        | NA       | Kaya et al. (2015d)  |
| **Vespa orientalis** |          |                |             |          |
|                   | 46.01      | 6.34           | 6.71        | NA       |                      |
| **Vespula**        |            |                |             |          |
| germanica          | 44.94      | 5.95           | 6.90        | NA       |                      |
| **Argynnia**       |            |                |             |          |
| pandora            | 44.89      | 6.53           | 6.62        | NA       | Kaya et al. (2015a)  |
| **Symperam**       |            |                |             |          |
| fonsecolmbii       | 44.91      | 6.45           | 6.48        |          |                      |
| **Brachymeria**    |            |                |             |          |
| portentosus        | 41.30      | NA             | 6.022       | 6.858    |                       |
| **Dociothsaurus**  |            |                |             |          |
| marocanus          | 42.35      | 5.64           | 4.63        | NA       | Erdogan and Kaya (2016) |
| **Cales**          |            |                |             |          |
| variabilis         | 45.44      | 6.31           | 6.23        | 7.29     | Kaya et al. (2015)   |
| **Dictius**        |            |                |             |          |
| verrucivorax       | 45.05      | 6.56           | 6.34        | 7.01     |                      |
| **Melanagryllus**  |            |                |             |          |
| desertas           | 48.90      | 6.88           | 6.08        | 8.04     |                      |
| **Paracyptera**    |            |                |             |          |
| labiana            | 46.10      | 6.41           | 6.25        | 7.38     |                      |

Fig. 3. XRD of (A) chitin and (B) chitosan extracted from five sources: cicada slough, silkworm chrysalis, mealworm, grasshopper and shrimp shells. Reprinted with permission (487329086712) from Carbohydrate Polymers (Luo et al., 2019), copyright 2019 Elsevier.
between 20.3 and 67% DW (Kaya et al., 2014; Kaya et al., 2016). Be
27.7% of the dry weight respectively. The chitin and chitosan content of
Dung beetle (Mingtang, 2004) was 17.32 and 14.48%, 13
11.3%, 3.90–8.40 and 78.33–83.33%, 12.70–14.20 and 75–83.37% and
28.7% of the dry weight respectively. The chitin and chitosan content of coleopteran
different insect species was found to be high, ranging from 94.3% to
99.3%. Previous reports have found that the solubility of mussel, oyster
shell, crab, pang scale, silver scale, prawn and conus shell chitin was
85.71%, 77.78%, 70.67%, 68%, 67.74%, 58.33% and 72.35%, respectiv
ately (Albaraayoe, Achiloun, & Hester, 2018; Mohan et al., 2019).
3.2. Solubility
The solubility (1% of aqueous acetic acid) of chitosan extracted from
different insect species was found to be high, ranging from 94.3% to
99.3%. Previous reports have found that the solubility of mussel, oyster
shell, crab, pang scale, silver scale, prawn and conus shell chitin was
85.71%, 77.78%, 70.67%, 68%, 67.74%, 58.33% and 72.35%, respectiv
ately (Albaraayoe, Achiloun, & Hester, 2018; Mohan et al., 2019).
3.3. Water binding capacity and fat binding capacity
Water binding capacity is the tendency of water to associate with
hydrophilic substances. Fat binding capacity is a measure of the amount
of oil absorbed per unit weight. The WBC and FBC of chitosan isolated
from a cicada, silkworm chrysalis, mealworm, and grasshopper were
noted to be 795–574%, 635–412%, 643–408%, and 594–275%, respec
the dissolution of chitin difficult (George & Roberts, 1992, pp.
249–267). Chitin is insoluble in many organic solvents, but chitosan
is substantially soluble in dilute acidic solutions with pH ≤ 6.0 (Chang,
Lin, Wu, & Tsai, 2015; Kumari, Annamarreddy, Abanti, & Rath, 2017;
Zargar, Asghari, & Dashni, 2015). The solubility of chitosan relies on the
temperature, the alkali concentration, the ratio of the chitin in alkali
solution, particle size, percentage of the degree of deacetylation (DD),
Mw, and biological origin (Hossain & Iqbal, 2014; Samar, El-Kalyoubi,
Khalaf, & Abd El-Razik, 2013). Based on the above factors, the solubility
of insect chitosan is similar to that of crustacean shells, and the high
solubility of insect chitosan should therefore be employed in many
useful applications in the future.
Table 4
Surface morphology (SEM analysis) of insect chitin and chitosan.

| Species                      | Surface morphology                  | Chitin Pore diameter | Chitosan Pore diameter | References                  |
|------------------------------|-------------------------------------|----------------------|------------------------|------------------------------|
| *Bradypterus (C.) sureyi*     | Nanofiber and nanopore              | 10 μm                | NA                     | Kabalak et al. (2020)        |
| *Gryllotalpa gryllotalpa*     | Nanofiber and nanopore              | 12–17 μm             | NA                     |                              |
| *Polyphila falk*              | Nanofiber and nanopore              | 4.5 μm               | NA                     |                              |
| *Omphius sp*                  | Nanofiber with porous surface       | 150–400 nm           | NA                     | Kaya et al., 2016a           |
| *Melolontha melolontha*       | Nanofiber with porous surface       | 185–400 nm           | NA                     | Kaya et al., 2014b, 2016b    |
| *Ranatra linearis*            | Nanofiber                           | NA                   | Nanofibre              | Kaya et al., 2014a           |
| *Anax imperator*              | Nanofiber                           |                      |                        |                              |
| *Hydrophilus piceus*          | Nanofiber                           |                      |                        |                              |
| *Notonecta glauca*            | Nanofiber                           |                      |                        |                              |
| *Agabus bipustulatus*         | Nanofiber                           |                      |                        |                              |
| *Leptinotarsa decemlineata*   | Nanofiber                           |                      |                        |                              |
| *Catharina molosus*           | Nanofiber                           |                      |                        |                              |
| *Silkworm chrysalis*          | Nanofiber                           |                      |                        |                              |
| *Grasshopper*                 | Nanofiber                           |                      |                        |                              |
| *Holotrichia parallelta*      | Rough and thick surface             | NA                   | NA                     | Liu et al. (2012)            |
| *Schistocerca gregaria*       | Nanofibers with pores               |                      |                        | Marei et al. (2016)          |
| *Apis mellifera*              | Nanofibers                           |                      |                        |                              |
| *Calosoma rugosa*             | Smooth surface with tiny pores      | NA                   | NA                     | Soon et al. (2018)           |
| *Periplaneta americana*       | Oval nanopores without nanofibers   | 230–510 nm           | NA                     | Kaya et al. (2015b)          |
| *Blaberus giganteus*          | Nanofibers and pores                | NA                   | NA                     | Kaya et al. (2017)           |
| *Hermetia illucens*           | Larvae                              | Porous surface       | NA                     | Wang et al. (2013)           |
|                              | Prepuca                             | Rough surface with no holes | NA                     |                              |
|                              | Puparium                           | Rough surface with irregular holes | NA |                              |
|                              | Adult                               | Rough and flocculent | NA                     |                              |
| *Hermetia illucens*           | Honeycomb structure and no porosity | NA                   | NA                     | Wasko et al. (2016)          |
| *Chrysonya megacephala*       | Nanofibers and with rarely distributed pores | NA | NA | Song et al. (2013) |
| *Cicada sloughs*              | Rougher morphology                  | NA                   | NA                     | Sajomsang and Gonil (2010)   |
| *Cicadatra atra*              | Nanofibers with nanopores           | NA                   | NA                     |                              |
| *Cicadatra hylalina*          | Nanofibrils and with rarely distributed pores | NA | NA |                              |
| *Cicadatra platyptera*        | Fibrous and porous                  | NA                   | NA                     |                              |
| *Cicadatra lodosi*            | Fibre bundles without pores         | NA                   | NA                     |                              |
| *Cicadatra mordogamensis*     | Fibre bundles without pores         | NA                   | NA                     |                              |
| *Cicadetta tibialis*          | Nanofibils and with rarely distributed pores | NA | NA |                              |
| *Honey bee*                   | Rougher morphology                  | NA                   | NA                     | Kaya et al. (2015d)          |
| *Wing*                        | Regular rough surface               | NA                   | NA                     |                              |
| *Head*                        | Highly fibrous and rarely porous    | NA                   | NA                     |                              |
| *Legs*                        | Very highly fibrous and rarely porous |                      |                        |                              |
| *Thorax*                      | Overlapped scales                   | NA                   | NA                     |                              |
| *Abdomen*                     | Only porous without fibers          | NA                   | NA                     |                              |
| *Vespa crabo*                 | Nanofibers and nanopores            | 100 and 200 nm       | NA                     | Kaya et al. (2015a)          |
| *Vespa orientalis*            | Nanofibers and nanopores            | 100 and 200 nm       | NA                     |                              |
| *Vespa germanica*             | Nanofibers and nanopores            | 100 and 200 nm       | NA                     |                              |
| *Vespa crabro*                | Nanofibils and pores                | NA                   | NA                     | Kaya et al. (2016c)          |
| *Argynnis pandora*            | Overlapping scales, smooth porous, tubular structures with big pores, plane area with no pores, rough surface | 20 μm | NA | Kaya et al. (2015a)          |
| *Ephestia kuehniella*         | Pores and parallel nanofibers       | 5.2 μm               | NA                     | Mehranian et al. (2017)      |
| *Silkworm chrysalides*        | Fine loosely united leaves           | NA                   | Porous structure       | Paulino et al. (2006)        |
| *Brachycerpes presentosus*    | Nanopores, thread-like fibrous      | 0.30–0.89 μm         | Big pores and fibres   | Ibitoye et al. (2018)        |
| *Greenbottle*                 | Porous with highly adherent nanofibers | 180–260 nm           | NA                     | Kaya et al. (2015)           |
| *Calliphus barbatus*          | Smooth surface                      | NA                   | NA                     | Kaya et al. (2015b)          |
| *Cedulas decorus*             | Nanofibers and nanopores            | NA                   | NA                     | Kaya et al. (2015c)          |
| *Pygmaphora cognata*          | Nanofibers and nanopores            | NA                   | NA                     |                              |
| *Oedipoda caerulescens*       | Nanofibers with no pores            | NA                   | NA                     |                              |
| *Oedipoda miniata*            | Nanofibers and nanopores            | NA                   | NA                     |                              |
| *Aiolopus strepens*           | Nanofibers and nanopores            | NA                   | NA                     |                              |
| *Aiolopus simulatrix*         | Nanofibers and nanopores            | NA                   | NA                     |                              |
| *Doroniella fraxi*            | Nanopores and nanofibers            | NA                   | NA                     |                              |

(continued on next page)
Table 4 (continued)

| Species               | Surface morphology          | Pore diameter | Chitosan | Pore diameter | References       |
|-----------------------|-----------------------------|---------------|----------|---------------|-----------------|
| Duroniella laticornis | Nanopores and nanofibres   | NA            | NA       | NA            | Marei et al. (2019) |
| Schistocerca gregaria | Fibrous structure           | NA            | NA       | NA            |                  |

Fig. 6. TGA curves for chitins from seven grasshopper species (a. Chitin from Ailopus simulatrix, b. Chitin from A. strepens, c. Chitin from Duroniella fracta, d. Chitin from D. laticornis, e. Chitin from Oedipoda miniata, f. Chitin from O. caerulescens, g. Chitin from Pyrgomorpha cognata and h. Commercial chitin). Reprinted with permission (4873291045484) from International Journal of Biological Macromolecules (Kaya et al., 2014), copyright 2014 Elsevier.
Table 5: Thermogravimetric analysis (TGA) of insect chitin and chitosan.

| Species                  | Chitin | Chitosan | References       |
|--------------------------|--------|----------|------------------|
|                          | First mass loss (°C) | Second mass loss (°C) | DTG<sub>max</sub> peak (°C) | First mass loss (°C) | Second mass loss (°C) | DTG<sub>max</sub> peak (°C) |
| Melolontha melolontha   | 4      | 78       | 380              | NA                | NA                | NA                |
| Ranatra linealis         | 6      | 78       | 393              | 9                 | 50                | 289               |
| Anax imperator           | 6      | 75       | 387              | 9                 | 87                | 295               |
| Hydrophilus piceus       | 5      | 73       | 386              | 3                 | 59                | 288               |
| Notonecta glauca         | 7      | 73       | 385              | 8                 | 61                | 308               |
| Agabus bipustulatus      | 5      | 71       | 384              | 6                 | 67                | 296               |
| Aelius aquaticus         | 5      | 71       | 350              | 8                 | 74                | 280               |
| Melolontha sp.           | 5.4    | 81.2     | 384.6            | NA                | NA                | NA                |
| Bradyprorus (C.) sureyi  | 5.2    | 72       | 382.4            | NA                | NA                | NA                |
| Gryllotalpa gryllotalpa  | 6      | 70       | 374.6            | NA                | NA                | NA                |
| Polyphylla fullo         | 5.9    | 73       | 374.7            | NA                | NA                | NA                |
| Lucanus cervus           | 6.6    | 70       | 379.9            | NA                | NA                | NA                |
| Omophlus sp.             | 3.6    | 78.8     | 385.3            | NA                | NA                | NA                |
| Leptinotarsa             | 4      | 74       | 379              | 5                 | 59                | 289               |
| decemlineata             | 3      | 48       | 307              | 5                 | 59                | 292               |
| Periplaneta americana    | 5      | 76       | 389              | NA                | NA                | NA                |
| Blaberus giganteus       | 6.44   | 71.69    | 401.7            | NA                | NA                | NA                |
| Adult                    | 5.96   | 71.37    | 374.1            | NA                | NA                | NA                |
| Hermetia illucens        | 4.42   | 69.48    | 372              | NA                | NA                | NA                |
| Larvae                   | 6.74   | 71.16    | 373              | NA                | NA                | NA                |
| Prepupa                  | 8.52   | 71.25    | 371              | NA                | NA                | NA                |
| Puparium                 | 7.5    | 73.31    | 372              | NA                | NA                | NA                |
| Adult                    | 7.5    | 73.31    | 372              | NA                | NA                | NA                |
| Hermetia illucens        | 5      | 70       | 363              | NA                | NA                | NA                |
| BSFE                     | 6      | 80       | 371              | NA                | NA                | NA                |
| Hermetia illucens        | 2      | 62       | 389              | NA                | NA                | NA                |
| Larvae                   | 3      | 63       | 387              | NA                | NA                | NA                |
| Cigada sloughs           | 7.3    | 66.4     | 362              | NA                | NA                | NA                |
| Cicada atra              | 4.54   | 83.75    | 411.50           | NA                | NA                | NA                |
| Cicadatru hyalina        | 5.47   | 66.78    | 412.70           | NA                | NA                | NA                |
| Cicadatru lodosi         | 4.41   | 83.94    | 411.70           | NA                | NA                | NA                |
| Cicadatru montogenetis   | 4.88   | 80.44    | 412.40           | NA                | NA                | NA                |
| Cicadatru platyperta     | 3.80   | 81.78    | 412.20           | NA                | NA                | NA                |
| Cicadatru tibialis       | 4.04   | 73.49    | 402.30           | NA                | NA                | NA                |
| Honeybee                 | 6      | 67       | 308              | NA                | NA                | NA                |
| Head                     | 4      | 56       | 360              | NA                | NA                | NA                |
| Thorax                   | 3      | 68       | 367              | NA                | NA                | NA                |
| Abdomen                  | 5      | 68       | 359              | NA                | NA                | NA                |
| Legs                     | 3      | 60       | 359              | NA                | NA                | NA                |
| Wings                    | 6      | 73       | 383              | NA                | NA                | NA                |
| Vespa crabo              | 6      | 83       | 385              | NA                | NA                | NA                |
| Vespa orientalis         | 6      | 76       | 385              | NA                | NA                | NA                |
| Vespa germanica          | 3.51   | 88.70    | 384.8            | NA                | NA                | NA                |
| Larvae                   | 2.7    | 69.9     | 381.7            | NA                | NA                | NA                |
| Pupa                     | 6.5    | 78.3     | 384.2            | NA                | NA                | NA                |
| Adult                    | 4.8    | 76.7     | 386.9            | NA                | NA                | NA                |
| Argennis pandora         | 4.9    | 82.2     | 389.6            | NA                | NA                | NA                |
| Other body parts         | 2.9    | 73.2     | 369.2            | NA                | NA                | NA                |
| Symperatum               | 4      | 77       | 386              | 5                 | 62                | 308               |
| fumicolumbi              | 4      | 82       | 383              | 7                 | 59                | 302               |
| Dociosaurus marocanus    | 3      | 80       | 386              | NA                | NA                | NA                |
| Adult                    | 5      | 87       | 388              | NA                | NA                | NA                |
| nymph                    | 5      | 94       | 385              | NA                | NA                | NA                |
| Cole viviablis           | 8      | 72       | 381              | 8                 | 61                | 296               |
| Decius verrucivorus      | 6      | 77       | 390              | 9                 | 57                | 305               |
| Melanoxyphus desertus    | 6      | 82       | 383              | NA                | NA                | NA                |
| Paracyptera labiata      | 5      | 78       | 382              | NA                | NA                | NA                |
| Callictus barbarus       | 6      | 74       | 381              | 8                 | 61                | 296               |
| Oedaleus decorus         | 6      | 77       | 384              | 9                 | 57                | 305               |
| Ailopus simulatrix       | 5      | 78       | 382              | NA                | NA                | NA                |
| Ailopus streps           | 6      | 74       | 381              | NA                | NA                | NA                |
| Durioella fructa         | 5      | 72       | 382              | NA                | NA                | NA                |
| Durioella laticornis     | 3      | 76       | 385              | NA                | NA                | NA                |
| Oedipoda miniata         | 5      | 77       | 384              | NA                | NA                | NA                |
| Oedipoda caeruleus       | 4      | 74       | 384              | NA                | NA                | NA                |
extracted from *Schistocerca gregaria*, *Apis mellifera*, and *Calosoma rugosa* were 516-307%, 511-304%, and 506-300%, respectively (N. H. Marei et al., 2016). The WBC and FBC of chitosan from crab (*Chionoecetes opilio*) legs range from 355% to 611% and 217%–403% (No, Lee, & Meyers, 2000). The WBC and FBC, therefore, could vary based on differences in the crystallinity of the products, the amount of salt-forming groups, deproteinization and demineralization processes (Knorr, 1982; Kumari et al., 2017).

Table 6

| Sources                          | Chemical shift (ppm) | References                          |
|---------------------------------|----------------------|-------------------------------------|
|                                 | C1       | C2       | C3       | C4       | C5       | C6       | C–O     | C–C     | G–C     | CH3     |             |
| Cicada sloughs chitin           | 104.2    | 55.3     | 73.5     | 83.3     | 75.8     | 61.0     | 173.8   | NA      | NA      | 23.0   | Sajomsang and Gonil (2010) |
| Silkworm pupa exuviae chitin    | 104.4    | 55.4     | 73.6     | 83.4     | 75.9     | 61.1     | 173.5   | NA      | NA      | 23.0   | Zhang et al. (2011)        |
| Beetle larvae cuticles chitin   | 104.4    | 55.7     | 74.0     | 83.6     | 76.1     | 61.5     | 174.3   | NA      | NA      | 23.0   | Majtán et al. (2007)       |
| Bumblebee cuticles chitin       | 103.9    | 54.9     | 73.1     | 82.7     | 75.5     | 60.6     | 173.3   | NA      | NA      | 22.3   | Paulino et al. (2006)      |
| Silkworm chrysalides chitin     | 104.5    | 55.6     | 73.8     | 83.5     | 76.1     | 61.4     | 173.3   | NA      | NA      | 23.2   | Song et al. (2013)         |
| Blowfly larvae chitosan         | 104.47   | 56.78    | 75.14    | 85.31    | 75.14    | 60.41    | 174.0   | NA      | NA      | 22.64  | Purkayastha and Sarkar (2020) |
| Black soldier fly chitin Imago  | 104.6    | 55.7     | 74.2     | 84.0     | 76.4     | 61.5     | 173.9   | NA      | NA      | 23.4   |                      |
| Pupae exuviae                   | 103.4    | 55.0     | 73.3     | 82.7     | 75.5     | 60.7     | 172.6   | NA      | NA      | 22.7   |                      |
| Silkworm chrysalides chitin     | 104.5    | 55.6     | 73.8     | 83.5     | 76.1     | 61.4     | NA      | NA      | NA      | 23.0   | Simionato et al. (2006)    |
| Silkworm chrysalides chitosan   | 105.3    | 57.9     | 75.8     | 82.3     | 75.8     | 61.1     | 174.0   | NA      | NA      | 23.0   |                      |
3.4. Ash and moisture content

It is necessary to quantify the ash content in chitin and chitosan before beginning the demineralization process, and it is important to evaluate its efficiency for the elimination of calcium carbonate. The demineralization process results in products containing 31%–36% ash (Kaya, Erdogan, et al., 2015). A high-value grade of chitosan should have <1% ash content (Nessa et al., 2010). The ash content of chitin and chitosan from fish (1.2% and 1.0%), shrimp (0.03%), crab (2.5%), conus shell (1.2%), honeybees (9.2%), beetles (2.0%, 2.20% and 0.50%), locusts (1.6%), cicada slough (0.03% and 11.3%), silkworms (0.05%), grasshoppers (0.89%), housefly larvae (0.13%), house crickets (1.0%) and Hermetia illucens (3.3, 5.6 and 19%) were measured (Caligiani et al., 2018; Ibitoye et al., 2018; Kumari et al., 2017; N. H.; Marei et al., 2016; Purkayastha & Sarkar, 2020; Sajomsang & Gonil, 2010; A.-J.; Zhang et al., 2011). Low ash content could be a reason for the superior...
solubility of chitosan (Kumar, Xavier, Lekshmi, Balange, & Gudipati, 2018). Furthermore, the moisture content can determine the performance of the powder when used in capsule/pill preparations. The moisture content of chitin and chitosan isolated from fish (13.8% and 3.0%), shrimp (0.0004%), crab (0.0048%), conus shell (6.5%), honeybee, beetles, locusts, cicada slough, silkworms, grasshoppers and house crickets were 17.6%, 8.8%, 14.1%, 7.12%, 0.18%, 0.19%, 1.8%, 8.7%, 4% and 3.33%, respectively (Kumari et al., 2017; Liu et al., 2012; Luo et al., 2019; N. H.; Marei et al., 2016; Mohan et al., 2019). Importantly, the moisture content of chitosan is not dependent on the Mw or the DD (Cho, No, & Meyers, 1998).

3.5. Molecular weight (Mw)

The Mw of commercial chitosan is between 100 and 1200 kDa (Li, Dunn, Grandmaison, & Goosen, 1992). The molecular weight of chitin and chitosan differs based on the source and the extraction methods used. The average viscosity Mw of chitin from honeybees and grasshopper larvae and adults is 738.806 kDa, 7.2 kDa, and 5.6 kDa, respectively (Draczynski, 2008; Erdogan & Kaya, 2016). The Mw of the Orthoptera chitin varied between 5.2 and 6.8 kDa (Kaya, Baran, & Karaarslan, 2015). The Mw of chitosan extracted from Colorado potato beetle adults (Kaya et al., 2014) and larvae, grasshoppers (Luo et al., 2019), Periplaneta americana, Hermetia illucens and Musca domestica (Ai, Wang, Yang, Zhu, & Lei, 2008; Jing et al., 2007) were 2.722 kDa, 2.676 kDa, 4.5 kDa, 3.779 kDa, 4.090 kDa, 3.975 kDa, 3.989 kDa, 230.3 kDa, 15 kDa, 426 kDa, and 63 kDa, respectively. High molecular weight is responsible for the poor solubility of chitosan in water and its high solution viscosity, which limits its use in the cosmetics, agriculture and food industries. The lower molecular weight chitosan from shrimp shells demonstrates higher antibacterial activity (Du, Zhao, Dai, & Yang, 2009), as does the low molecular weight (25 kDa) chitin extracted from conus shell (Mohan et al., 2019). Chitosan has a moderate molecular weight and demonstrates higher anti-cholesterol activity (Kara & Stevens, 2002). The Mw of insect chitin and chitosan could be determined by viscometry methods (Draczynski, 2008; Erdogan & Kaya, 2016; Kaya et al., 2014; M. W.; Kim, Song, Han, et al., 2017) and high-performance liquid chromatography. The diverse Mw of chitin can be used in many useful ways. The low Mw chitin and chitosan from shrimp and insects have excellent antiseptic and anticancer properties useful for drug development.

3.6. Degree of deacetylation (DD)

The DD of chitin and chitosan is the significant parameter influencing the biological, physicochemical, and mechanical properties dependent on the method of extraction (Khan, Peh, & Ch‘ing, 2002). The DD of chitosan was 94.9% in Catharsius molossus, 89%, 96% (Ma et al., 2015) and 95% in locusts, honeybees and beetles (N. H. Marei et al., 2016), 81.06% in Zophobas morio (Soon et al., 2018), 91.86% in Periplaneta americana, 42.47% in Hermetia illucens (Khayrova et al., 2019), and 83% and 90.3% in housefly larvae (Ai et al., 2008; A.-J.; Zhang et al., 2011); the DD of chitin was 133%, 86%, 121%, 120%, 117% and 86% in Ranatra linears, Anaz imperator, Hydrophilus piceus, Notoneeta glauca, Agabus bipustulatus and Asellus aquaticus, respectively (Kaya et al., 2014). Several methods have been developed for the determination of DD in chitin and chitosan from insects. Among them, the potentiometric titration method (Ma et al., 2015), the conductometric titration method (Khayrova et al., 2019), the acid-base titration method (A.-J. Zhang et al., 2011) and the FT-IR (Kaya et al., 2014) are effective for perfectly soluble materials. The DD of chitosan from fish, shrimp, and crab shells was 75%, 78% and 70%, respectively (Kumari et al., 2017). Previous studies have suggested that a higher DD is a significant development of chitin that can be used in scaffolds and implantations in the biomedical field (Akpan, Ghenebor, & Adeosun, 2018).

Fig. 8. 3D scatter plot of structural characterization studies (XRD, EA, TGA and NMR analysis) in insect chitin and chitosan.
4. Structural characterization

The structural characterization of insect chitin and chitosan was determined by X-ray diffraction, elemental analysis, Fourier transform infrared spectroscopy, scanning electron microscopy, thermogravimetric analysis, and nuclear magnetic resonance spectroscopy.

4.1. Crystalline properties

The CrI values of chitin and chitosan are significant in determining their potential application areas (Aranaz et al., 2009), as they depend on their crystalline and amorphous nature. This could be detected using X-ray diffraction. Nevertheless, the crystalline nature also represents the purity and size of the crystals in the biopolymer. As noted in previous studies, a low crystalline index (CrI %) was obtained in chitin from *Hermetia illucens* at the larval (33.05%) and prepupal (35.14%) stages. However, the puparium (68.4%) and adult (87.92%) stages of the same species have also had high CrI recorded (Caligiani et al., 2018). High molarity (2 M) NaOH during the deproteinization process has been found to increase the amorphous nature and decrease the crystallites of insect chitin. Furthermore, the surface morphology of the obtained chitin had a lower CrI with an amorphous region with a porous surface compared to the higher CrI that had a rough and irregular surface (Table 2). According to Park et al. (2010), the CrI was measured as the ratio between the area of the crystalline contribution and the total area. Similarly, the total XRD peaks obtained from *Agabus bipustulatus* and *Brachytrupes portentosus* showed 7 and 10 distinct peaks at 2θ with the highest CrI of 90.6% and 88.02% (Ibitoye et al., 2018; Kaya et al., 2014). This finding also indicates the impurity of the chitin obtained from *B. portentosus* using N-6.02%. CrI values of chitosan from cicada slough, silkworm chrysalises, mealworms, grasshoppers and shrimp shells were observed to be 64.8%, 32.9%, 51.9%, 50.1% and 49.1%, respectively, and the crystallinity indices of shrimp shells, mealworms and grasshopper chitosan were similar (Luo et al., 2019) (Fig. 3). The chitosan extracted from crab and squilla exhibited two characteristic crystalline peaks at 2θ = 10.3° and 19.2° and 20 = 10.2° and 19.5°, which were slightly shifted to a higher diffraction angle and showed semi-crystalline chitosan (Anand et al., 2014). *Vespa crabro, Vespa orientalis, Vespula germanica, Argynnis Pandora, Ailopus simulatrix* (Kaya, Baran, & Karaarşlan, 2015; Kaya et al., 2016) exhibited 6 crystalline peaks and a CrI between 69 and 76%. Moreover, a high number of XRD peaks attributed to impurities (6.6–6.9% N-factor) have been found to be present in...
insect chitin, which influences the degradation of the polysaccharides with DTG at ≈383–386 °C. In addition, the chitosan showed 3 variant peaks demonstrated from Schistocerca gregaria and Brachyrurus portentosus in thread-like fibrous structures with a crystal size of ≥7.21 nm to 0.12 μm (Ibitoye et al., 2018; N. H.; Marei et al., 2016), which is large compared to other insect chitosan reported to date. Furthermore, all published literature reports the crystalline properties of insect chitin to be in the range of ≥60–90% Crl, although these numbers would differ based on the alkaline and acidification used in the extraction process. The chitin with the higher Crl value obtained from insects is an alternative chitin source that can be used in the biomedical field. The XRD patterns of the chitin and chitosan extracted from all insects species are also quite similar (Fig. 8).

4.2. Elemental analysis (EA)

Elemental analysis of chitin from different types of insects, including the carbon, nitrogen, hydrogen and carbon-nitrogen ratio are shown in Table 3. The percentage of C atoms from chitin originating from various insects ranged from 32.09% to 48.90%. The N content of chitin is a significant indicator of its purity, and the N content of pure (acetylated) chitin has been found to be 6.89%. Nitrogen content >6.89% (Liu et al., 2012; Majtán et al., 2007; Sajomsang & Gonil, 2010) shows that protein residues may still be present in the chitin sample, though nitrogen content <6.89% suggests that inorganic materials have not been completely removed. The N% value of chitin from Melolontha melolontha, Periplaneta americana, Vespa crabo, Argynnis pandora, and Symmetry fonssonii was measured to be 6.72%, 6.69%, 6.85%, 6.62%, and 6.83%, respectively (Kaya, Bağraçık, et al., 2015; Kaya, Baublys, et al., 2014; Kaya, Bulut, et al., 2016). Additionally, the EA results for the chitin from crab was 6.03%, 42.9% and 6.55%; from crayfish was 6.09%, 42.88%, 6.02%; and from shrimp was 6.17%, 43.2%, 6.42% (Kaya, Baran, & Karaarslan, 2015). The N% values of the chitin from insects from different orders were very close to the theoretical value. The above studies show that chitin obtained from insects is of high purity. In this context, the elemental composition of the chitin and chitosan extracted from all insect species is similar (Fig. 8).

4.3. Fourier transform infrared spectroscopy

FT-IR spectroscopy is generally used for the identification of organic samples (Dukor, Story, & Marcott, 1999). There are three crystalline forms of chitin, which are alpha, beta and gamma, but there is little information about the gamma form (Jang, Kong, Jeong, Lee, & Lee, 2004). FT-IR spectra is helpful for differentiating between the alpha and the beta form using the presence or absence of the amide I band. In the alpha form, the amide I band divides into two bands at approximately 1650 and 1620 cm⁻¹ (Wang et al., 2013), while in the beta form, there is only one amide I band in the 1656 cm⁻¹ region. Beta chitin is found in squid pens (Jang et al., 2004), and alpha chitin is found in the order Arthropoda (Al-Sagheer et al., 2009; Sajomsang & Gonil, 2010). In the FT-IR spectra of the chitin and chitosan extracted from various insects (Fig. 4), such as Holotrichia paralela (Liu et al., 2012), Zophobas morio (Shin et al., 2019), Periplaneta americana (Kaya, Baran, & Karaarslan, 2015), Hermetia illucens (Wasko et al., 2016), and Apis mellifera (Kaya, Lelesius, et al., 2015), the amide I band is split at 1654 cm⁻¹, 1663 and 1618 cm⁻¹, 1647 and 1654 cm⁻¹, 1654 and 1621 cm⁻¹, 1654, 1617 and 1550 cm⁻¹, and 1656 cm⁻¹, respectively. The FT-IR spectra of the amide I band of the chitosan extracted from squilla, crab, corns shelf, krill, lobster and shrimp is split at 1643 cm⁻¹, 1634 cm⁻¹, 1625 cm⁻¹, 1628 cm⁻¹ and 1667 cm⁻¹, respectively (Anand et al., 2014; Mohan et al., 2019; Sayari et al., 2016; Sriniwasan et al., 2018; Wang et al., 2013). These results show that the chitin and chitosan isolated from crustacean shell waste and insects are in the α-form.

4.4. Scanning electron microscopy

Scanning electron microscopy is an instrumental technique for the visual confirmation of the morphology and physical state of the surface of chitin. The surface morphology of insect chitin and chitosan differs according to the organisms from which they originate. Generally, insect chitin and chitosan may be classified into the following surface morphologies (Table 4): (I) nanofibre and nanopore, (II) nanofibre, (III) nanopores without nanofibres, (IV) nanofibres without nanopores, (V) smooth surface, and (VI) rough surface. Crickets (Kabaliak et al., 2020), grasshoppers (Kaya, Bağraçık, et al., 2015), Orthoptera species (Kaya, Baran, & Karaarslan, 2015) (Fig. 5) and house cricket chitin (Ibitoye et al., 2018) show both nanofibre and nanopore structures. Aquatic bugs, water scavenger beetles, desert locust (Kaya et al., 2014) and Colorado potato beetle chitosan (N. H. Marei et al., 2016) have a nanofibrous structure. A few reports have shown that cockroach and black soldier fly chitin had nanopores without nanofibres and nanofibres (Kaya et al., 2014) without nanopore structures (Wasko et al., 2016). In addition, the chitin from Zophobas morio and Holotrichia paralela and the chitosan from Cathartius molassus had smooth and rough surface morphologies. In this context, Anand et al. (2014) reported that sponge and cauliflower leaf-like morphology was observed in crab and squilla chitin. The SEM analysis of conus chitin showed a microfibrillar crystalline structure and porosity (Mohan et al., 2019). The tightly arranged fibres were also observed in the chitin obtained from krill, shrimp and lobster shell (Al-Sagheer et al., 2009; Wang et al., 2013). Furthermore, SEM analysis of the chitin and chitosan surface morphologies of P. monodon showed microfibril and porous structures (Sriniwasan et al., 2018). Surface morphology is one of the vital properties that determines the effective use/application of chitin and chitosan. The nanofibre and nanopore forms of chitin and chitosan could be used in textiles, food and therapeutic applications (Aranaz et al., 2009; Synowiecki & Al-Khateeb, 2003).

4.5. Thermogravimetric analysis

The thermal stability of the chitin and chitosan from insects is measured in the mass losses found at two steps (Table 5; Fig. 6). The loss at the first step is attributed to the evaporation of water from the chitin and chitosan molecules, and the loss at the second step represents the degradation of the chitin and chitosan units (Ofem, 2015). Anand et al. (2014) reported in the TGA analysis of chitosan from crab and squilla that mass loss occurred three stages; the first mass loss occurred below 100 °C, followed by a second mass loss (252 °C, 269 °C, and 213 °C) and a third mass loss (367 °C, 384 °C and 350 °C). Ladchumanandavism, da Rocha, Belarmino, and Galv (2012) demonstrated that decomposition occurs in the ranges of 50–100 °C and 400–500 °C for shrimp and crab chitin. For all the chitin samples from various insects, the first mass loss was noted to be between 2% and 8.52%, while the second mass loss ranged from 48% to 94% (Ladchumanandavism et al., 2012). The maximum degradation temperatures (DTGmax) of chitin extracted from different insect orders ranged between 307 °C and 412.40 °C (Kaya, Baublys, et al., 2014; Kaya, Lelesius, et al., 2015; Kaya et al., 2016; Mol et al., 2018; Sajomsang & Gonil, 2010). The above findings concluded that insect chitin molecules could disintegrate at higher temperatures than chitosan molecules. This variance could be due to the N-acetylated polymer units of chitin molecules that are more stable than the amine polymer units of chitosan (Paulino et al., 2006). These results indicated that insect chitin molecules are more stable than insect chitosan units. Additionally, the thermal stability of chitin and chitosan extracted from all insect species is similar (Fig. 8).

4.6. Nuclear magnetic resonance spectroscopy

NMR spectroscopy is the most potent structural elucidation technique for organic compounds, and it functions using a magnetic field and
radiofrequency pulses transmitted at a particular resonant frequency to detect the signal of specific nuclei, including $^{1}H$, $^{31}P$, or $^{13}C$, in the region of interest (Mandal, 2007). The solid-state $^{13}C$ NMR is useful for the structural characterization of carbohydrate polymers such as chitin and chitosan without damaging the samples. $^{13}C$ CP/MAS NMR spectroscopy could be used to determine the assignments of carbon chemical shifts of chitin and chitosan from various insect sources, as shown in Table 6. The $^{13}C$ CP/MAS NMR spectra of the cicada slough chitin spectrum contains eight well-defined peaks of C1–C6, CH3, and C=O carbons, which are detected by a chemical shift ranging from 20 to 190 ppm (Fig. 7). The C1–C6 carbons displayed a chemical shift ranging from 50 to 110 ppm, while the methyl carbon and the carbonyl carbon showed a chemical shift of 23 ppm and 174 ppm, respectively (Sajomsang & Gonil, 2010). The $^{13}C$ CP/MAS NMR spectrum of the chitosan from blowfly larvae, Chrysomya megacephala, consists of seven well-defined peaks of C1 (δ 104.47), C2 (δ 56.78), C3 (δ 75.14), C4 (δ 85.31 and 80.97), C5 (δ 75.14), C6 (δ 60.41) and CH3 (δ 22.64) and identified a weak methyl resonance (δ 22.64) representing a relatively high degree of acetylation (C. Song et al., 2013). This study indicated that highly deacetylated chitin and chitosan had more biological properties than less deacetylated chitin and chitosan (Heux, Brunnerotto, Desbrieres, Versalli, & Rinaudo, 2000). Moreover, the chemical shifts in the NMR from the chitin and chitosan extracted from all insect species are similar (Fig. 8).

5. Biological activities

Insect chitin and chitosan have a broad spectrum of biological activities, such as antioxidant effects and antibacterial effects with substantial rheological properties, which could be used in the food industry to enhance food safety, shelf-life and quality control.

5.1. Antioxidant activity

Free radicals are produced by abnormal metabolic processes and cause extensive damage to living organisms, which may result in several diseases, such as cancer, inflammation, and neurodegenerative diseases (Halliwell, 2011; Moskovitz, Yim, & Chock, 2002). Commonly, free radicals are effectively removed by antioxidant enzymes in the body. Generally, naturally derived compounds have been used to treat free radical-mediated harmful effects in biological systems. Numerous studies have examined the antioxidant activities of chitin and chitosan from insects (Ai et al., 2008; Kayá, Bitim, et al., 2015; Kayá, Bulut, et al., 2016; C.; Song et al., 2013; Torres-Castillo et al., 2015; S.-J.; Wu et al., 2013). Chitosan from the adult Colorado potato beetle with low MW has been reported to have a higher DPPH radical scavenging action at a concentration of 5 mg/mL, but chitosan obtained at the larvae stage of the same species displayed only 33.05% of the scavenging action with MW. However, these chitosan showed similar action against the ferric ion reducing test. Furthermore, this study stated that a higher degree of acetylation (DA) had high antioxidant action, while the DA of the adult and larval Colorado potato beetle was 82% and 76%, respectively (Kaya et al., 2014). Additionally, no FRAP action was recorded in chitosan and colloidal chitin polymers derived from DNA fragmentation chitin from commercial shrimp shell (Kidibu, Santos-Moriano, Plou, & Fernande-z-Lobato, 2020); nonetheless, hydrolysis of the polymers improved FRAP action between 77% and >90%. In comparison with this result, chitosan derived from C. barbarus and O. decorus displayed lower reactions of 33.51%, and 33.26% in DPPH scavenging activity at a concentration of 5 mg/mL (Kaya, Bitim, et al., 2015). This action was less efficient compared to the housefly Musca domestica, which displayed the highest DPPH scavenging effect of 57.1% at a low concentration of 0.5 mg/mL (Ai et al., 2008). Furthermore, this outcome suggested that these two species, which can be catastrophic to food crops, could possibly be considered as a potential source of chitin and chitosan to be used in the food/feed industry for its antimicrobial properties.

5.2. Antibacterial activity

Recent findings have confirmed that insect chitin and chitosan possess significant antibacterial activity. In a few reports, shrimp and crab shell chitosan demonstrated better action against Gram-negative microbes than Gram-positive organisms (Chung et al., 2004). The possible mechanism for this difference could be the hydrolysis of peptidoglycan due to interactions between the positively charged chitosan molecules and the negatively charged microbial cell membranes (Fig. 9). This interaction leads to the collapse of the cell membranes, escape of the intracellular components, and ultimately, to cell death (Chien, Yen, & Mau, 2016). However, chitosan from two grasshopper species, C. barbarus and O. decorus, showed a potential effect against both gram-positive and gram-negative microbes compared to standard antibiotics. The gram-positive bacteria were L. garvieae, S. agalactiae, L. monocytogenes, and B. subtilis, and the gram-negative bacteria, such as Y. enterocolitica, V. alginolyticus, and S. enteritidis showed minimal bactericidal concentrations (MBCs) of 0.32 mg/mL and 0.16 mg/mL for the chitosan derived from both grasshopper species (Kaya, Erdogan, et al., 2015). Similarly, chitoooligosaccharide extracted from the cicada Cryptotympana atrata displayed maximum zones of inhibition against B. subtilis, S. aureus, and E. coli of 9.52 mm, 12.64 mm, and 10.79 mm, respectively. These chitoooligosaccharides confirm the linkage of the β-1, 4-linked 2-amino-2-deoxy-d-glucopyranose (GlcN) and 2-acetamido-2-deoxy-d-glucopyranose (GlcNAc) (S.-J. Wu et al., 2013). This linkage has been found to be similar to that of COS from crustacean chitin (Polybius henslowii crab), which displayed a better inhibition against the fungi Cryptophora parasitica at a concentration from 0.0125 to 0.1 mg/mL (Avelelas et al., 2019). However, chitoooligosaccharides from Clantis bilineata indicated significant inhibitory action against B. subtilis, which was found to be similar to that of commercial chitosan (S. Wu, 2011). Furthermore, 4% deacetylated chitosan from T. molitor mealworm beetle larvae did not show any inhibitory effect against S. aureus, B. cereus, L. monocytogenes, or E. coli, but increasing the chitosan concentration to 8% resulted in 1–2 mm of inhibition. The crystallinity index (Cr I) value of T. molitor chitosan was 58.11% compared to that of fish waste chitosan, which ranged from 36 to 71% (Kumari et al., 2017). A chitin film developed from B. giganteus cockroach wing and the dorsal pronotum region limited biofilm formation by A. baumannii and S. sonnei bacteria. Furthermore, a 7-day incubation of the fungal strain A. nigra on the surface of the chitin film demonstrated 7.6 × 10^9 spores, but the wing chitin film had 4.26 × 10^6 spores. A. nigra spores (Kaya et al., 2017). Nevertheless, ciprofloxacin loaded nanoparticles developed from chitosan derived from insects such as beetles (Calosoma rugosus) and honeybee (Apis mellifica) exoskeletons displayed similar inhibition against Methicillin-resistant Staphylococcus aureus with an MIC of 0.14 μg/mL (N. Marei et al., 2019). This finding demonstrates that the antibacterial effects of insect chitosan can also be used as active edible packaging in food applications (Hamed et al., 2016; R.; Muzzarelli & Muzzarelli, 2005).

5.3. Rheological properties

Rheology is the study of flow and deformation of food materials and is a vital tool for characterizing the fundamental material properties, such as processing, handling, quality control, storage and sensory evaluation of food ingredients (Kutz, 2007). During food production and processing, several materials are often in liquid form. Polysaccharides are comprised of chain conformations and produce bio-macromolecular aggregates when scattering in the presence of water molecules, which could be due to the intermolecular hydrogen bonding. In most cases, biopolymers have pseudoplastic or non-Newtonian properties that aid in their applications in food production and pharmaceuticals. However, flow property is profoundly influenced by polysaccharide structural arrangements, the pH of the medium, the temperature applied to the system and the ionic concentrations of the external matter. Chitosan
derived from cicada slough, silkworm chrysalises, mealworms, and grasshoppers (prepared as a 2% solution with 1% aqueous acetic acid) exhibited a high shear rate and shear-thinning behaviour compared to shrimp shell chitosan with a sweeping decline in viscosity. Similarly, chitosan with a higher Mw possesses higher viscosity; for instance, shrimp shell chitosan, which has a Mw of 1.620 × 10^5 Da, showed high viscosity, and cicada slough, which possess a low Mw of 3.777 × 10^3 Da, had low viscosity (Luo et al., 2019). However, these two factors are highly influenced by the degree of acetylation (DD) and are decreased by the degree of deacetylation (DDA) (Liu et al., 2012). Alternately, polymers expressing shear-thinning behaviours demonstrate pseudoplastic fluid/non-Newtonian characteristic features in food applications. Decreasing the NaOH concentration to less than 50% in chitosan extraction reduces the DDA reaction and increasing the percent NaOH decreases viscosity. Similar results were obtained in the chitosan derived from houssely larvae extracted using 50% (w/v) at 125 °C for 4 h, which exhibited ~79% DDA with ~347 mPa.s viscosity and 60% NaOH in the extraction process had ~82% DDA with ~250 mPa.s viscosity (A.-J. Zhang et al., 2011). Additionally, 1 M NaOH at 80 °C with a varied time of 39, 44, 49, 54, 59, and 64 h showed a significant reduction in the intrinsic viscosity ranges from 30.6 to 18.9 η from chitin obtained from honeybees (Draczyński, 2008). Furthermore, the quality of housefly larvae chitosan was equivalent to food-grade chitosan according to the Chinese Fishery Trade Standard SC/T3403-2004. Therefore, orthogonal experiments or optimization of multiple parameters in insect chitosan extraction could provide appropriate storage modulus (G') and loss modulus (G'') in food applications (Nishinari, 1997). Nevertheless, shrimp shell chitosan expressed more G'' with high viscous properties, and as a result of this characteristic, crustacean-derived chitosan is directly used in many food applications. In addition, insect chitosan solutions donate non-covalent cross-linking at a low level, which might be utilized as low viscosity chitosan (X. Zhang & Waymouth, 2017). In the future, the lower viscosity of insect chitosan could be used as a thickening and suspending material for the food industry.

5.4. Wound healing

Engineering skin substitutes provides a prospective source of advanced therapy to combat acute and chronic skin wounds. The wound healing process involves multiple consecutive reaction pathways, including haemostasis, aggregation, cell multiplication, and regeneration (Goldberg & Diegelmann, 2010). This process contains various cell types, including the extracellular matrix and cytokine mediators active in healing. The wound healing mechanisms of chitin and chitosan from insects are shown in Fig. 10. Recently, skin substitutes using biomaterials from natural materials have been used as wound dressings. For example, desert locust (Schistocerca gregaria) chitosan was tested for the wound remodelling process in a mouse model. A 9 mm wound created on the mouse’s back displayed potential wound closure when treated with locust chitosan (N. H. Marei et al., 2016). This chitosan reduced the inflammatory necrosis on the skin cells after 5 days of treatment for up to 14 days. A similar healing process has been found with shrimp chitosan, but a higher count of dermis active angiogenesis was found using seeded locust chitosan. It was reported that 1–2% of chitosan from P. niloticus (freshwater crab) increased the thickness of the epidermis in wounded rats compared to a high concentration (3%) of chitosan applied to the wound (Amer & Attia, 2020). Furthermore, researchers stated that chitosan consists of glycan derivatives that might act as macrophage stimulating agents as well as initiating cytokine production from the macrophages. These two reactions amplify the wound healing process in the early phase (Ueno et al., 1999), and insect chitosan may therefore be a promising natural wound healing material.

5.5. Anti-tumour

Chitin and chitosan derived from insects have shown substantial anti-tumour activities. The in vitro inhibitory effect of chitosan from housefly Musca domestica larvae displayed 50.8% and 52.9% action against HeLa and S-180 tumour cells at 1 mg/mL in an MTT assay. Furthermore, this chitosan could chelate ferrous ions in vitro, which is considered an effective pro-oxidant found in the food system that induces cell proliferation. It was noted that native and inoculated larvae of Musca domestica extract demonstrated antitumour action against the human colon cancer cell line CT26 with an inhibition rate of 62–89% at 500 and 1000 μg/mL of extract. However, this wholesome extract also showed the presence of peptidoglycan as an active ingredient and exhibited antitumour action (Hou, Shi, Zhai, & Le, 2007). In contrast, lower concentrations (400 μg/mL and 200 μg/mL) of chitosan from P. longipalpis (shrimp) displayed >50% cytotoxic activity against Human larynx carcinoma (Hep2) cells and Human embryo rhabdomyosarcoma (RD) cells (Ganesan et al., 2020). Nevertheless, chitosan nanoparticles (CNPs) demonstrated competitive action at low concentrations. For example, 80 and 100 μg/mL of CNP from Musca domestica, Lucilia sericata, and Chrysomya albiceps exhibited productive anticancer activity against human liver carcinoma (HepG-2) and human colon carcinoma (HCT-116) cell lines. These CNPs reported an IC50 value of 37.3–74.3 μg/mL, with the most potent inhibition recorded from C. albiceps CNP (Hasbullah, 2019). Hence, insect chitosan could serve as alternative therapeutic agents for the treatment of tumours.

5.6. Anti-ageing

Ageing is a natural process that affects most biological activities and seems to be a consequence of the cumulative action of various types of stressors. Evidence shows that oxidative stress from ROS, telomere attrition, a decline in DNA repair and protein turnover systems serve as significant causes of ageing (Kirkwood, 2005; Vijg & Campisi, 2008). Oxidative stress is caused by the disparity between ROS production and ROS removal in the biosystem, which leads to oxidative injury to cells and tissues and alterations in their morphology and function, resulting in ageing and age-related disorders, such as cognitive deficits and Parkinson’s disease (Shan et al., 2009). The anti-ageing activities of chitin and chitosan from insects are rarely reported. Wu et al. (2016) reported that different concentrations of water-soluble chitosan of Clanis bilimnetae larva skin were intragastrically administered to D-gal-induced mice at 42 days. The results indicated that the administration of chitosan significantly increased superoxide dismutase (SOD) and glutathione peroxidase (GPs) and decreased malondialdehyde (MDA) in the brains and sera of the mice. This finding suggests that Clanis bilimnetae chitosan could be used as an effective antioxidant an anti-ageing medicine. In comparison with insect chitin, crustacean chitin, chitin-nanofibrils and chitin-hyaluronan nanoparticles have been reported to increase the creation of fibroblasts, inhibit IL-8 and TNF-α release, and trigger antioxidant enzyme release from the skin layer in addition to their skin-hydrating properties (Morganti et al., 2013). However, further innovative mechanisms are required to explain the anti-ageing activity of insect chitin and chitosan.

5.7. Hypolipidaemic activity

Hyperlipidaemia, characterized by high levels of fats in the blood and the impairment of lipid metabolism, is a major cause of atherosclerosis and subsequent related cardiovascular diseases (Ahmad & Beg, 2013; Natar-Boggan et al., 2015; Prasad & Kalra, 1993). In recent years, many studies have focused on the reduction of serum lipid levels and the absorption of fat in the intestinal tract to reduce chronic diseases (A.-J. Zhang et al., 2011). Hence, the antihyperlipidaemic activity of many bioactive components from natural materials such as polysaccharides are novel possible hyperlipidaemic agents (Knopp, 1999). Insect chitosan and its derivatives have a lowering effect on plasma cholesterol, which plays a vital role in the prevention and treatment of cardiovascular disease, although minimal investigations have examined these
effects of insect chitin and chitosan (Anraku et al., 2010; Lamiaa & Barral, 2011). Xia et al. (2013) stated that chitooligosaccharides (COS) from Clavis bilineata fed rats at 6 weeks had the ability to prevent increases in body weight and to lower plasma triglycerol (TG), total cholesterol (TC), and plasma low-density lipoprotein cholesterol (LDL-C) levels. These results showed that insect COS could be used as an alternative hypolipidaemic drugs. Other chitin sources, such as fungal, crustaceans and sponges have also been reported to have hypolipidaemic actions. These chitins downregulated adipogenesis and adipocyte-specific gene promoters by modulating adiponectin monophosphate-activated protein kinase (AMPK) and aquaporin-7 (Kong, Kim, Bak, Byun, & Kim, 2011). Further investigation is required to examine the AMPK signalling pathway to confirm the anti-hypolipidaemic activity of insect chitin.

5.8. Industrial application

Chitosan is a biodegradable cationic biopolymer that could aid in the decrease of metal pollutants from industrial effluents through the adsorption and chelation of particles through productive electrostatic activity (Evans, Davida, MacRae, & Amirbahman, 2002). This action could act in the agglutination of colloidal particles. The use of chitin and chitosan from shrimp as an adsorbent agent has been widely investigated for the removal of azo dyes from the textile industry (Duarte, Ferreira, Marvao, & Rocha, 2002; Szyguła, Guibal, Ruiz, & Sastre, 2008). The chitin and chitosan from silkworm chrysalides at concentrations of 50 mg/L and 21.3 mg/L reduced the amount of the anthraquinone dye and residual aluminium (Al) in textile industry effluents by 6 and 70 h. The study indicated that adsorption quality is higher in insect chitosan than in insect chitin (Julliana I Simionato, Paulino, Garcia, & Nozaki, 2006; Julliana Isabelle Simionato et al., 2014).

6. Shortcomings and possible technical solutions

Extracting chitin from insect biomass is undoubtedly more challenging compared to marine sources. Even though green technologies or process optimization may lead to high quantity products, it is evident that this could only be accomplished through extensive research. Research related to understanding the feasibility of the techniques and variances in proximate composition and processing conditions should continue to be explored in this field. For example, untreated larvae, including blanched and dried larvae, did not exhibit chitin due to their high-fat content (3–20%) (Khavraeva et al., 2019). However, at this stage, the use of phosphoric acid in chitin extraction might not be useful due to the hydrophobic repulsion that occurs on the cell wall of the insect (Mba, Kansei, Vial, Rougerie, & Genot, 2019). Similarly, the amount of pigment in the insect cell could influence chitin extraction. It was reported that melanin covalently binds to chitin at the pupae or late-stage of insects and blocks the extraction of chitin using organic acids (H$_3$PO$_4$). Therefore, these challenges should be rectified using depigmentation processes or by choosing non-pigmented insects for chitin extraction. These challenges again necessitate multiple-steps for chitin extraction, and in order to scale-up and lessen the extraction procedures, it is required to develop novel/innovative technologies. Recently, an electrochemical technique was identified to minimize the multiple-downstream methods used for the removal of lipids, proteins and pigments from marine organism-based chitin (Nowacki et al., 2020). Two primary steps involved in this method use catholyte and anolyte treatments in two chambers within the same system. It was engaged at a high pH (12.5) of the electro-alkali in the cathode chamber (at 70 °C for 16 V, 1.5 A), which lysed the cell walls and partially degraded the lipids, proteins and pigments. It was reported that the chitinous skeleton was removed from the interlayer spaces of Cirripedae sp (black coral) during this step. Moreover, deep eutectic solvents (DES), also known as novel ionic liquids that are comprised of hydrogen bond donors (HBDs) and acceptors (HBA), could be suitable for insect chitin extraction. Some common HBDs are betaine, HCl, ChCl, etc., and HBAs such as urea, ethylene glycol and glycerol have been used at minimum temperatures of 50–90 °C. HBDs and HBAs have been applied to skimmed black soldier flies (Hermetia illucens) and showed efficient results (Zhou et al., 2019). Chitin extracted by DES was found to have a high purity (74–91.345) and yield (12.71–26%) compared to the conventional acid/alkali method (purity 91% and yield 6.5%). It was observed that the best efficiency of deproteinization was obtained by using highly acidic solvent in a HBD at a high temperature (80–90 °C) in the extraction system, which leads to increased protein removal of approximately 3%–10% (Zhou et al., 2019). However, DES-integrated with microwaves showed better deproteinization efficiency (88–93% rate of removal) in shrimp chitin (D. Zhao et al., 2019; Y. Zhao et al., 2010). Therefore, the integrated method using microwave, autoclaving, and enzymatic treatments would be appropriate to simplify the chitin extraction process from insects. In addition, designing a suitable electrochemical system with the involvement of electrolysis would be useful for scaling up the quantity of chitin obtainable from insects.

7. Challenges and opportunities

Globally, industrial chitin/chitosan producers rely upon marine-derived sources for its production. Major commercial plants for chitin/chitosan production are located in various countries, including Europe (https://mealfoodeurope.com), USA (https://tidalvisionusa.com), India (http://thahirachemicals.com/profile.html), and France (https://chitosanlab.com). Most of these industries use the exoskeletons of shrimp, crab, squid bone, or fungi, etc., for large scale chitin production. Therefore, various strategies are required to extend the commercialization of insect chitin/chitosan conversion at industrial levels. A few industries, such as Sfyl®, utilize Hermetia illucens larvae for high-quality chitin/chitosan (http://sfylproteins.com/sfyl-products/) production. This demonstrates the lack of technology transfer in the scaling up of insect chitin, which needs to be addressed. Some challenges involved in the extraction of chitin from insect sources are (1) Insect collection: The gathering of catastrophic species (locusts, crickets, termites, etc.) would require specific techniques, but they are not consistently available throughout the year. Similarly, a suitable processing method should be adopted to retain the chitin proportion until its extraction, which leads to additional requirements ideal for various species. Therefore, the cost of conversion of biomass into a useable form for extraction could exceed unit operational costs. (2) Extraction: Process optimization is crucial for insect chitin extraction. While increased alkaline (NaOH/KOH) concentrations could negatively affect the total quantity of chitin extracted, the same condition favours a deproteinization process. Similarly, few concentrated acids (H$_3$PO$_4$) showed hydroporphic repulsion on the insect exoskeleton, but some (HCl and H$_2$SO$_4$) are found to hydrolyse chitin. Therefore, identifying efficient solvent mixtures appropriate for insect species are required for mass production. Therefore, technological innovations are essential to deviate from the conventional downstream processes using single components. Positively: Insects have been used as a meal in Europe that has received significant attention due to its high protein content (https://mealfoodeurope.com/en/tecnologia/). Meanwhile, industries are breeding and insects for high quality and quantity. Cricket flies as baking ingredients (https://thecricketbakery.com/) and mealworms in snacks (https://www.diewurmfarm.at) are a few examples of cultured insects in food applications. Meanwhile, these insects are consumed as wholesome food/feed in various parts of the world and obtained approved by the European Commission as a novel food (EC, Regulation (EC) No 1069/2009), Regulation (EU) 2016/759 (EC), demonstrating that insect chitin could have direct applications in the food system without any regulatory issues.

8. Future perspectives

Globally, the market for chitin and chitosan is growing steadily. Due
to the pandemic disease COVID-19, there is an increase in the demand for biopolymer materials for healthcare, personal care, packaging, and coating materials, and this emerging situation has increased the demand for biomaterials for biomedical, food, and pharmaceutical applications. The projected statistics show the market size for chitin/chitosan will grow up to 162.7 thousand MT, mainly derived from 15.6% of chitosan with a growth rate of 17.6% in the following years (Newswire, 2019). Furthermore, in-depth research has been conducted on chitin/chitosan applications, including scaffolds in tissue engineering (wound healing), drug release encapsulation, food packaging, coating, 3D scaffolds, and hydrogels from marine-invertebrate waste, with less focus on insect chitin. Therefore, studies of 3D chitin and chitosan from insect shells are needed for biomedical applications. Additionally, food security issues are another alarming problem due to the devastation of food-crops by locust (grasshopper) waves. Though agricultural scientists are working on measures for controlling these pests, converting waste into valorization would be a significant technique for its prevention.

Therefore, the future direction of research should focus on the destruction of catastrophic species into a value-added product that could replace the existing biopolymers and increase the opportunities in this field. Further studies are required to optimize the production process for higher yield using electrochemical methods or integrated approaches such as ultrasonication and microwave. Innovative insect rearing methods would also produce a constant supply of specific species/stages of insects for industrial needs. Methods with cost-effective and straightforward synthesis approaches could be required for large-scale production of insect chitin. Therefore, up-scaling efficiency, insect species selectivity, and stability in real-time applications need to be explored.

Declaration of competing interest

The authors declare no conflict of interest.

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