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Development Prospect and Preparation Technology of Edible Oil From Microalgae

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Microalgae are a valuable and rich source of oil. By the means of using microalgae oil as raw materials or additives for edible oil will improve the nutritional and edible value of the latter. This paper compares the lipid content and fatty acid composition of microalgae and traditional oil crops, elaborates the lipid synthesis mechanism of microalgae cells and the strategies of promoting lipid accumulation both from environmental pressure and from molecular pressure, summarizes the methods of breaking microalgae cell walls and extracting oil, analyzes the nutritional value, toxicity, antioxidant, and economic feasibility issues to be considered in microalgae edible oil. It is hoped that this study could provide theoretical basis for the actual production of microalgae edible oil, and provide inspiration for the research on the synthesis, accumulation, extraction and preparation of microalgae oils.

Keywords: microalgae oil, edible oil, omega-3 PUFAs, oil accumulation, oil extraction

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INTRODUCTION

As population growth and the development of the biodiesel industry, traditional sources of edible oil have faced the challenge. Food researchers have focused on microalgae, an effective, sustainable and promising source of food and fuel supplies. Using microalgae as a source of edible oil save agricultural resources. They grow in non-cultivated land like seawater, salt water and even wastewater. With only sunlight and some nutrients, the autotrophic microalgae used for oil production can grow in large quantities in indoor photoreactors (Raheem et al., 2015; Chen et al., 2018). Microalgae are able to produce large amounts of oil per unit area, and their biomass productivity is 10 times higher than that of plankton and much higher than that of terrestrial biomass. Their fatty acid composition is the same as that of plant-derived oil, mainly C16 and C18 fatty acids. Some species are also rich in valuable ω-3 polyunsaturated fatty acids (PUFAs), which have antioxidant activity and cardiovascular benefits (Li X. et al., 2019; Suparmaniam et al., 2019; Katiyar and Arora, 2020). As a result, microalgae oil is considered as a functional oil with great commercial potential, and has the potential to be as a raw material for ordinary edible oil or as an added nutrient enhancer.

In recent years, there have been many studies on the production of biodiesel from microalgae (Ahmad et al., 2011b; Salam et al., 2016; Skorupskaite et al., 2016; Faried et al., 2017; Raheem et al., 2018; Sivaramakrishnan and Incharoensakdi, 2018; Srinuanpan et al., 2018a,b; Mofijur et al., 2019; Shomal et al., 2019). Biodiesel is a mixture of fatty acid alkyl esters (FAAE), most commonly methacrylate (FAME) or ethyl ester (FAEE), obtaining by alcoholysis of triacylglycerol (TAG)

from microalgae oil (Halim et al., 2012). Studies are concentrated on the selection of oil-producing algae species (Nwokoagbara et al., 2015; Yu et al., 2015; Arroussi et al., 2017; Sadvakasova et al., 2019), fatty acid composition and fuel characteristics (Islam et al., 2017; Piloto-Rodríguez et al., 2017), upstream cultivation of microalgae for biodiesel production and downstream processing (Wang et al., 2016; Zhu et al., 2017, 2019; Kadir et al., 2018; Mathimani and Mallick, 2018; Tan et al., 2018; Goh et al., 2019; Yew et al., 2019; Yin et al., 2020), and analysis of economic feasibility and energy potential (Shin et al., 2018; Arcigni et al., 2019; Sun J. et al., 2019). Though there are few researches and evaluations on the production of microalgae edible oil.

This article studied the development prospect and preparation technology of edible oil from microalgae, compared the microalgae oil with vegetable oil to highlight the advantages of the former in the lipid productivity and fatty acid composition, introduced the microalgae cell lipid synthesis pathways and the method of the promoting intracellular lipid accumulation based on environmental pressure and molecular strategies, summarized the mechanical and non-mechanical methods to break microalgae cells, and analyzed organic solvent, ionic liquid, supercritical fluid, enzyme-assisted water and some other oil extraction methods, finally looked forward to the prospect of edible oils from microalgae.

THE OIL CONTENT AND FATTY ACID COMPOSITION OF MICROALGAE

Overview of Microalgae Lipids

Microalgae, one of the most primitive forms of plants, live in water systems. They are very small single-celled or simple multicellular photosynthetic microorganisms, having no leaves or roots, sizes <400 μm, and diameters usually 1–30 μm (Mata et al., 2010; Vassilev and Vassileva, 2016). Most algae are eukaryotes. The nuclei of eukaryotic microalgae are similar to those of higher plants, with intracellular organelles including chloroplasts for photosynthesis, endoplasmic reticulum (ER), golgi apparatus, mitochondria and vacuoles. The main biochemical components of microalgae are lipids, proteins, polysaccharides and nucleic acids (Sajjadi et al., 2018).

Microalgae cells typically contain 30–80% lipids (Japar et al., 2017; Deshmukh et al., 2019). Lipids are separate from polar and non-polar. Polar lipids (phospholipids and glycolipids etc.) are important components of cell membranes, while non-polar lipids (triglycerides and sterols etc.) are mostly used for intracellular energy storage (D'Alessandro and Antoniosi Filho, 2016). Microalgae have accumulated large amounts of non-polar lipid TAG, which are tiny droplets of three identical or different fatty acids molecules attached to glycerol. Moreover, in terms of fatty acid composition, microalga oil contains PUFAs with multiple double bonds. PUFAs are divided into different classes inclusive of ω-3 PUFAs and ω-6 PUFAs, which are, respectively, synthesized from linoleic acid and linolenic acid, and are essential for human health (Lorgeril and Salen, 2012; Jang and Park, 2019). The ω-3 PUFAs have received extensive attention owing to their physiological effects such as antioxidant activity, immune regulation, inflammation reduction, and prevention of neurological and cardiovascular diseases, especially the two most important α-linolenic acid derivatives, docosahexaenoic acid (DHA, C22: 6ω-3) and eicosapentaenoic acid (EPA, C20: 5ω-3) (Wu et al., 2015; Elagizi et al., 2018; Luo et al., 2018; Ferguson et al., 2019; Heshmati et al., 2019; Oikonomou et al., 2019; Ahmmed et al., 2020). It has been proved in animal experiments that intaking of diets rich in DHA and EPA in appropriate forms will enhance cognitive ability (Wen et al., 2016; Zhou et al., 2018), regulate metabolism (Asztalos et al., 2016; Bargut et al., 2017; Hussein et al., 2019) and have anti-inflammatory effects (Sierra et al., 2008; Calder, 2015; Alfaddagh et al., 2018). DHA is abundant in the brain and retina and plays a vital role in promoting the development of the brain and retina and maintaining membrane fluidity to ensure normal function (Bazan, 2007; Echeverría et al., 2017; Sun et al., 2017). DHA also ameliorates vascular health, participates in reducing inflammatory responses, prevents cancer, and protects the body from disease (Jung et al., 2013; Ozkan et al., 2016; Yum et al., 2016). EPA plays a beneficial role in fat metabolism, and prevents obesity, atherosclerosis and other diseases by improving fat metabolism (Laiglesia et al., 2016; Ding et al., 2017; Zhang et al., 2017).

Microalgae Lipids and Plant Oil

Compared with traditional vegetable oil, microalgae have become a potential new source of edible oil as its advantages in oil content and fatty acid composition. **Table 1** (Balke and Diosady, 2000; Ogunniyi, 2006; Ong et al., 2011; Wang et al., 2012; Rondanini et al., 2014; Mukherjee and Ghosh, 2017; Dorni et al., 2018; Beyzi et al., 2019; Dehghan et al., 2019; Ebrahimian et al., 2019; Hossain et al., 2019; Xie et al., 2019, 2020; Liu et al., 2020; Tamagno et al., 2020) lists the oil content and fatty acid composition of some conventional oil crops. In general, the oil content of plant-derived crops is 20–60%, and the oil yield is generally less than 300 gal oil/acre (Sajjadi et al., 2018). Besides castor oil contain special up to about 87.0% of ricinoleic acid [C20:0 (OH)], palm oil and coconut oil, respectively, contain quite amount of palmitic acid (C16:0) and lauric acid (C12:0), most plant oil containing unsaturated fatty acids 70%, unsaturated fatty acids primarily oleic acid, linoleic acid, linolenic acid, and 20 carbon olefine acid. In addition to flaxseed oil, mustard oil and double-low rapeseed oil, vegetable oil contain relatively low ratios of ω-3 PUFAs, especially safflower oil, where the ratio of ω-6 PUFAs to ω-3 PUFAs reaches 631.59 (Ferreira et al., 2019).

The lipid content of microalgae varies according to different algae species and growth periods, usually between 20 and 50% biomass, and come up to 70% under certain culture conditions. Lipid productivity rather than lipid content is generally accepted as an indicator for evaluating the oil-producing performance of microalgae. Lipid content is the concentration of lipids in microalgae cells, regardless of the production of biomass; and lipid productivity depends on the production of biomass, and refers to the accumulation of lipids in cells in the total biomass produced. Microalgae with low lipid content have higher lipid productivity in the main, for instance, *Chlorella* sp. with only about 30% lipid content and lipid productivity exceeding

TABLE 1 | The oil content and fatty acid composition of traditional vegetable oil.

Species	Soybean (Dorni et al., 2018)	Rapeseed (Beyzi et al., 2019)	Peanut (Dorni et al., 2018)	Castor (Mukherjee and Ghosh, 2017)	Linen (Liu et al., 2020)	Sunflower (Dorni et al., 2018)	Olive (Dehghan et al., 2019)	Palm (Dorni et al., 2018)	Mustard (Dorni et al., 2018)	Cottonseed (Dorni et al., 2018)	Corn (Dorni et al., 2018)	Coconut (Dorni et al., 2018)	Safflower (Dorni et al., 2018)
Gal oil/acre (Sajjadi et al., 2018)	46	122	109	145	49	98	124	23	59	33	18	276	80
Fat content %	~20 (Tamagno et al., 2020)	45–50 (Hossain et al., 2019)	45.9–55.4 (Wang et al., 2012)	46–55 (Ogunniyi, 2006)	37–40 (Xie et al., 2020)	35–50 (Ebrahimian et al., 2019)	35–46 (Rondanini et al., 2014)	46–50 (Ong et al., 2011)	31 (Balke and Diosady, 2000)	28.24–44.05 (Liu et al., 2020)	4–5	65–75	32–40 (Ebrahimian et al., 2019)
Palmitic acid (C16:0)	11.67 ± 0.48	4.24–6.00	10.46 ± 0.97	1.99 ± 0.01	5.17 ± 0.05	6.43 ± 0.36	14.05 ± 1.14	39.67 ± 0.79	2.19 ± 0.21	23.40 ± 0.23	12.94	9.26 ± 0.39	6.24 ± 0.53
Stearic acid (C18:0)	3.87 ± 0.82	0.36–2.41	3.37 ± 0.27	1.14 ± 0.01	5.36 ± 0.08	3.69 ± 0.22	3.41 ± 0.30	4.23 ± 0.11	1.17 ± 0.06	2.79 ± 0.12	2.12	2.97 ± 0.23	2.35 ± 0.16
Oleic acid (C18:1)	24.77 ± 2.41	53.95–60.98	53.77 ± 3.52	3.68 ± 0.12	26.90 ± 0.20	25.92 ± 2.23	69.58 ± 2.68	43.36 ± 0.61	10.16 ± 0.75	17.82 ± 0.18	31.97	7.24 ± 0.25	13.8 ± 0.62
Linoleic acid (C18:2)	54.17 ± 1.72	20.42–25.02	26.96 ± 2.42	4.74 ± 0.07	14.43 ± 0.09	62.69 ± 2.20	11.12 ± 1.70	11.23 ± 0.46	15.58 ± 0.43	51.81 ± 0.66	48.97	1.90 ± 0.14	76.58 ± 1.10
Linolenic acid (C18:3)	5.16 ± 0.77	8.74–9.56	nd	nd	46.70 ± 0.25	nd	0.97 ± 0.07	0.30 ± 0.05	11.70 ± 0.57	0.35 ± 0.00	0.76	nd	0.13 ± 0.03
Arachidic acid (C20:0)	0.37 ± 0.04	0.7	1.42 ± 0.13	nd	nd	0.32 ± 0.09	nd	nd	0.98 ± 0.03	0.42 ± 0.05	0.68	nd	0.26 ± 0.06
Eicosenoic acid (C20:1)	nd	2.82	nd	nd	nd	nd	nd	nd	5.48 ± 0.36	nd	nd	nd	0.17 ± 0.02
Others	–	1.93[a]	–	88.80 ± 0.16[b]	–	–	–	–	51.18 ± 1.31[a]	–	–	–	–
SFA	15.90 ± 0.65	15.85–20.50	19.27 ± 1.52	nd	nd	11.39 ± 0.46	nd	44.84 ± 0.90	5.73 ± 0.29	28.17 ± 0.47	16.60	90.84 ± 0.33	9.19 ± 0.58
MUFA	24.77 ± 2.41	58.22–60.27	53.77 ± 3.52	nd	nd	25.92 ± 2.23	nd	43.62 ± 0.61	66.98 ± 0.78	19.66 ± 0.19	33.67	7.24 ± 0.25	14.04 ± 0.60
PUFA	59.33 ± 2.08	19.35–24.06	26.96 ± 2.42	nd	nd	62.69 ± 2.20	nd	11.54 ± 0.47	27.28 ± 0.64	52.16 ± 0.66	49.74	1.90 ± 0.14	76.78 ± 1.15
PUFA/SFA	3.73	nd	1.40	nd	nd	5.51	nd	0.26	4.76	1.85	3.00	0.02	8.39
ω-6/ω-3 PUFAs	10.50	1.6–2.5 (Xie et al., 2019)	–	–	nd	–	nd	37.43	1.33	146.7	64.15	–	631.59

[a]*Erucic acid (C22:1);* [b]*Ricinoleic acid (C20:0(OH)); nd, not determine. SFA, saturated fatty acids; MUFA, monounsaturated fatty acids; PUFA, polyunsaturated fatty acids.*

100 mg/L/day (Kiran et al., 2014). On account of the high lipid productivity of microalgae, the oil production of microalgae is able to achieve 6,000–15,000 gal oil/acre, far over that of vegetable oil (Sajjadi et al., 2018). **Table 2** (Chisti, 2007; Bogen et al., 2013; Japar et al., 2017; Sajjadi et al., 2018; Shuba and Kifle, 2018; Deshmukh et al., 2019; Ferreira et al., 2019; Menegazzo and Fonseca, 2019). In terms of fatty acid composition, the content of PUFAs in microalgae is 20–60%. Some algal species, for example, *Phaeodactylum tricornutum*, *Botryococcus braunii*, and *Dunaliella salina*, contain DHA and EPA. Nevertheless, many vegetable oil contains only the precursor of both, α-linolenic acid, which has low conversion efficiency in human body (Gómez-Cortés and Camiña, 2019).

LIPID SYNTHESIS AND ACCUMULATION IN MICROALGAE

Metabolic Pathways for Microalgae Lipid Synthesis

The biosynthesis pathways of fatty acids and TAG in microalgae are similar to those in higher plants, including two stages of *de novo* synthesis of fatty acids and subsequent glyceride assembly (**Supplementary Figure S1**). The fatty acid synthesis begins in the chloroplasts in microalgae. Microalgae fix carbon dioxide into glycero-3-phosphate (G-3-P) through photosynthesis, and further convert it into acetyl coenzyme A (acetyl-CoA) through glycolysis, which is the direct precursor of fatty acid synthesis. In addition, the citric acid produced by sugar metabolism through the tricarboxylic acid (TCA) cycle in the mitochondria is also converted to acetyl-CoA under the action of ATP: citric acid cleavage synthase (ACL). In the cytoplasm, acetyl-CoA is activated by acetyl-CoA carboxylase (ACC) to generate malonyl coenzyme A (malonyl-CoA), which can be used for fatty acid elongation in the ER membrane. The conversion of acetyl-CoA to malonyl-CoA is the first step of fatty acid synthesis, which requires ATP. After the synthesis of malonyl-CoA in chloroplast, it is transferred to acyl carrier protein (ACP), and then goes through a four-step cycle of condensation, reduction, dehydration, and reduction (each cycle extends two carbon atoms), finally results in C16-ACP and C18-ACP. The elongation of C2 unit of each carbon chain demands 1 ATP and 2 NADPH molecules. On the one hand, acyl-ACP continues to synthesize lipids in chloroplast, which is considered as the prokaryotic pathway of lipid metabolism. On the other hand, acyl-ACP is dissolved into free fatty acids (FFA) under the action of fatty acyl-ACP thioesterase (FAT), FFA combine with the coenzyme A in the cytoplasm to regenerate the acyl-CoA, enter the ER with G-3-P through Kennedy pathway to generate lyso-phosphatidic acid (LPA), phosphatidic acid (PA), diacylglycerol (DAG) in turn, and become TAG in the end. There is a certain relationship between DAG, TAG, and membrane lipids (ML), this is eukaryotic way of lipid metabolism (Bellou et al., 2014; Arora et al., 2018; Mohan et al., 2019; Ran et al., 2019; Arif et al., 2020).

The ω-3 PUFAs are synthesized by both plastid and ER in the presence of specific elongases and desaturatase enzymes (Bellou et al., 2014). Synthesis of microalgae PUFAs begins with oleic acid, which is achieved by carbon chain elongation and desaturation of linoleic acid and linolenic acid (**Supplementary Figure S2**). In the traditionalω-3 PUFAs synthesis process, the eicosatetraenoic acid is generated by the Δ6 desaturation enzyme and elongation enzyme, and this process could also be completed by the Δ8 substitution process with the assistance of the Δ9 elongation enzyme and the Δ8 desaturation enzyme. Next, EPA is synthesized by the eicosatetraenoic acid under the action of Δ5 desaturation enzyme, and then DHA is synthesized by EPA under the Δ5 elongation enzyme and Δ4 desaturation enzyme (Guschina and Harwood, 2006; Gong et al., 2014; Hamilton et al., 2014).

Different substrates lead to different lipids synthesis of plant plasmids and ER. Plastid synthesis takes acyl-ACP as the substrates, and ER takes acyl-CoA as the substrates. Thus, it is possible to determine whether the synthesis site of glycerides is plastid or ER according to whether the fatty acyl group occupying the sn-2 position is 16 or 18 carbonyl groups. Unfortunately, reaction-related enzymes of microalgae have not been studied in detail, and the prokaryotic and eukaryotic pathways mentioned above may not be fully applicable to microalgae (Klok et al., 2014). Moreover, studies have identified at least six genes in the Chlamydomonas genome that presumably encode DGAT, and microalgae lipid synthesis may be much more complex than plant lipid synthesis for the reason of the wider substrate specificity of proteins (Liu and Benning, 2013). With the study of genome sequencing of microalgae, the differences between microalgae and plant lipid biosynthesis is more and more obvious. For example, the synthesis of TAG in microalgae usually takes place in photosynthetic cells, while that in plants mainly occurs in developing embryos and fruits. TAG synthesis of microalgae showed regionalized characteristics. Plasma membrane lipids are synthesized by plasmids, while extracellular lipids are synthesized by ER only. Due to the synthesis of DHA and EPA by microalgae cells, some aspects of desaturation and elongation of fatty acids are different from that of plants. Microalgae and plants have different fatty acyl groups, and the overall subcellular tissues of glyceride metabolism are different (Merchant et al., 2012; Liu and Benning, 2013; Klok et al., 2014; Zienkiewicz et al., 2016). Consequently, the lipid metabolism pathway of microalgae needs to be further studied.

Strategies of Microalgae Lipid Accumulation

When algae are subjected to environmental stress and modified at the molecular level, lipid metabolism is affected and accumulates (**Supplementary Figure S3**). Thus, microalgae can obtain high lipid content by changing the external culture conditions like strong light, extreme temperature, high salt, chemical induction, plant hormone regulation, and co-culture by using the unique symbiotic relationship between bacteria, fungi, and microalgae. Different stress factors mainly induce lipid accumulation in microalgae by changing ROS balance, carbon flow conversion and stress hormone levels, and regulating lipid synthesis and conversion (Chung et al., 2017; Sajjadi et al., 2018; Goh et al., 2019; Ran et al., 2019; Sun X. et al., 2019; Yew et al., 2019;

TABLE 2 | Lipid content and fatty acids composition of different microalgae species.

Microalgae species	Oil content (%) (Sajjadi et al., 2018)	Lipid productivity (mg/L/day) (Mata et al., 2010; Kiran et al., 2014)	Palmitic acid (C16:0)	Stearic acid (C18:0)	Oleic acid (C18:1)	Linoleic acid (C18:2)	Linolenic acid (C18:3)	EPA (C20:5)	DHA (C22:6)	SFA	MUFA	PUFA
Bacillariophyceae												
Phaeodactylum tricornutum (Deshmukh et al., 2019)	18–57	44.8	11.3	0.4	2.8	1.5	1.4	28.4	–	20.2	25.3	49.8
Thalassiosira weissflogii (Sajjadi et al., 2018)	5–20	nd	32.4	1.5	9.8	–	0.3	14.1	–	nd	nd	nd
Skeletonema costatum (Sajjadi et al., 2018)	13–51	17.4	7.25	1.15	1.55	1.4	0.5	20.8	3.5	nd	nd	nd
Chaetoceros muelleri (Sajjadi et al., 2018)	13–24	21.8	24.6	2.06	1.81	1.0	0.9	5.53	0.68	nd	nd	nd
Chlorophyceae												
Botryococcus braunii (Ferreira et al., 2019)	25–75 (Chisti, 2007)	5.5	9.36	1.27	59.55	7.00	11.20	2.23	3.38	11.69	63.10	25.21
Botryococcus terribilis (Ferreira et al., 2019)	49.00 (Menegazzo and Fonseca, 2019)	nd	6.27	1.28	75.19	3.09	5.20	1.24	2.26	10.60	77.03	12.37
Chlorella salina (Deshmukh et al., 2019)	11 (Menegazzo and Fonseca, 2019)	nd	21.5	7.83	14.39	10.88	29.75	–	–	29.34	18.52	40.63
Desmodesmus sp. (Ferreira et al., 2019)	6.5–9.1 (Ferreira et al., 2019)	nd	18.91	14.66	27.97	12.37	1.61	–	–	13.98	28.43	57.59
Desmodesmus brasiliensis (Ferreira et al., 2019)	17.99 (Menegazzo and Fonseca, 2019)	nd	26.19	7.05	15.49	14.62	20.24	–	–	36.94	26.56	36.50
Dunaliella primolecta (Sajjadi et al., 2018)	23	nd	23.9	1.2	11.4	6.6	39.9	–	1	nd	nd	nd
Dunaliella tertiolecta (Sajjadi et al., 2018)	11–16	60.6–69.8	26.1	4.05	8.6	22.9	41.4	0.4	–	nd	nd	nd
Dunaliella salina (Deshmukh et al., 2019)	6–25	116.0	18.2	0	4.56	13.24	30.26	–	–	18.71	5.54	58.9
Dunaliella sp. (Deshmukh et al., 2019)	22 (Menegazzo and Fonseca, 2019)	33.5	9.19	4.27	22.51	3.84	44.31	–	–	13.47	24.74	48.15
Monoraphidium contortum (Deshmukh et al., 2019)	22.2 (Bogen et al., 2013)	nd	20.9	0.3	17.3	10.5	23.9	–	–	24.4	18.1	56.3
Nannochloris sp. (Sajjadi et al., 2018)	20–56	60.9–76.5	18	1.84	37.5	11.6	13.5	0.03	0.4	nd	nd	nd
Nannochloropsis sp. (Ferreira et al., 2019)	31–68 (Chisti, 2007)	54.8	31.58	3.54	34.48	4.97	–	–	–	nd	nd	nd

(Continued)

TABLE 2 | Continued

Microalgae species	Oil content (%) (Sajjadi et al., 2018)	Lipid productivity (mg/L/day) (Mata et al., 2010; Kiran et al., 2014)	Palmitic acid (C16:0)	Stearic acid (C18:0)	Oleic acid (C18:1)	Linoleic acid (C18:2)	Linolenic acid (C18:3)	EPA (C20:5)	DHA (C22:6)	SFA	MUFA	PUFA
Nannochloropsis oculata (Deshmukh et al., 2019)	22–29	84.0–142.0	20.5	1.8	4.1	2.2	0.9	29.7	–	10.4	39.5	36.9
Scenedesmus obliquus (Ferreira et al., 2019)	30–50	nd	27.39	11.88	32.08	9.08	8.11	–	–	nd	nd	nd
Scenedesmus sp. (Deshmukh et al., 2019)	17–24	40.8–53.9	15.62	2.97	15.23	7.00	22.99	–	–	18.59	26.86	30.00
Ankistrodesmus sp. (*Deshmukhet al*., 2019)	11.48–31	nd	16.24	7.18	17.66	8.8	28.68	–	–	23.43	23.27	37.16
Ankistrodesmus falcatus (Deshmukh et al., 2019)	16.49 (Menegazzo and Fonseca, 2019)	49.58 ± 5.74	30.23	2.72	24.79	2.0	26.86	–	–	34.02	25.26	28.8
Chlamydomonas reinhardtii (Sajjadi et al., 2018; Menegazzo and Fonseca, 2019)	21	nd	17.8	6.85	32.4	6.58	16.0	–	–	28.18	22.88	32.07
Chlamydomonas sp. (Deshmukh et al., 2019)	22.7	nd	50.77	11.54	13.77	3.93	2.76	–	–	63.92	14.05	6.69
Eustigmatophyceae												
Chlorella sp. (Ferreira et al., 2019)	28–53	42.1	19.03	2.35	48.21	1.29	1.54	–	–	26.27	70.90	12.37
Chlorella vulgaris (Deshmukh et al., 2019)	41–58	11.2–40.0	35.77	11.35	12.55	7.36	13.54	–	–	25.08	22.69	52.23
Chlorella protothecoides (Deshmukh et al., 2019)	40–60	1214	13.42	3.4	58.94	19.86	–	–	–	19.79	58.94	19.86
Chlorella emersonii (Deshmukh et al., 2019)	23–63	10.3–50.0	14.75	9.8	17.01	9.04	29.32	–	–	24.55	17.01	38.37
Rhodophyceae												
Porphyridium cruentum (Deshmukh et al., 2019)	9–14 (Shuba and Kifle, 2018)	34.8	28.6	0.8	2.0	8.2	0.7	21.1	–	31.1	5.0	40.8
Prymnesiophyceae												
Pavlova salina (Deshmukh et al., 2019)	12–30	49.4	15.1	1.0	3.8	1.5	2.2	19.1	–	30.7	34.2	29.4
Cryptophyceae												
Chroomonas salina (Deshmukh et al., 2019)	12–14.5	nd	13.5	3.0	5.2	1.2	10.8	12.9	–	10.7	7.5	65.5
Conjugatophyceae												
Schizochytrium sp. (Sajjadi et al., 2018; Menegazzo and Fonseca, 2019)	50–77	nd	18.8	3.5	53.4	14.9	–	–	–	18.59	26.86	30.00

nd, not determine. SFA, saturated fatty acids; MUFA, monounsaturated fatty acids; PUFA, polyunsaturated fatty acids.

Zhao et al., 2019). One of the most common and effective means is nutritional stress, specially nitrogen stress (Breuer et al., 2015). On the basis of nutritional stress, a two-stage culture method is developed. In the first stage, microalgae achieve high biomass concentration in nutrient-rich culture, and after harvesting biomass, in the second stage, microalgae attain lipid accumulation under environmental stress (Aziz et al., 2020). Furthermore, genetically engineering microalgae by over-expression, heterologous expression, gene knockout, and inserting certain genes to increase the supply of acetyl-CoA and NADPH precursor substances, and controlling signaling pathways by transcription factor can make the balance transfer in the direction of lipid synthesis, thereby enhance the synthesis of fatty acids and TAG, inhibit lipid β-oxidation and hydrolysis, and reduce starch, sterol and other competition way (Singh et al., 2016; Tandon and Jin, 2017; Arora et al., 2018; Park et al., 2019; Arif et al., 2020).

For biodiesel, it is more meaningful to increase the content of microalgae TAG, because the existence of PUFAs is not conducive to the stability of fuel. However, as edible oil, the development and utilization of PUFAs should also be considered. In the early stage of nitrogen depletion, starch accumulates; while in the late depletion, lipid accumulation. Under the appropriate environment, the growth curve of microalgae consist of four periods, namely stagnation, logarithmic, linear and stable. The stable stage is of high lipid content (Aziz et al., 2020). Different lipids also accumulate at different times and under different conditions: PUFAs need to be produced by low illumination at the beginning of the stationary period, while TAG will be produced by high illumination at the end of the stationary period (Gifuni et al., 2019). Studies have shown that the establishment of a reasonable carbon and nitrogen supply strategy, the provision of sufficient dissolved oxygen in the medium, the control of light, radiation and the addition of chemical substances are conducive to the accumulation of PUFAs (Ling et al., 2015; Chua and Schenk, 2017; Chen and Yang, 2018; Hu H. et al., 2019).

EXTRACTION AND PREPARATION OF EDIBLE MICROALGAE OIL

Cell Disruption

Despite the many advantages of microalgae, the commercial application of oil meets with formidable challenges for the high cost of downstream processing including the process of cell wall breaking and oil extraction. Microalgae cells are small and have a dense cell wall. Conventional mechanical pressing methods, which are widely applied to extracting vegetable oil, are not suitable for microalga. Direct solvent extraction or other methods to extract oil are also less efficient. Microalgae cell crushing methods are comprised of mechanical and non-mechanical methods (**Supplementary Figure S4**) (Grimi et al., 2014; Günerken et al., 2015; Lee et al., 2017; Onumaegbu et al., 2018; Goh et al., 2019; Hu Y. et al., 2019; Li X. et al., 2019; Mathimani and Mallick, 2019; bin Azmi et al., 2020; Katiyar and Arora, 2020; Sankaran et al., 2020). Commonly used mechanical cells broken pretreatment methods are mainly physical methods, such as ball mill, homogeneous and cavitation, but non-mechanical methods include chemical method and biological method. When selecting different methods, energy consumption and oil extraction effect should be considered. Mechanical methods have high energy consumption, high-pressure homogenization, high-speed homogenization and ultrasonic methods may cause lipid oxidation and seriously affect product quality, while chemical and biological methods have low energy consumption but the possibility of pollution, and may produce some by-products (Grimi et al., 2014; Günerken et al., 2015; Onumaegbu et al., 2018; Menegazzo and Fonseca, 2019). Combining complementary advantages of two or more methods with each other often gains better oil extraction effect (Duan et al., 2018; Phong et al., 2018). After cell fragmentation, fatty acid composition may also be affected. Previous studies have shown that mechanically broken cells increase the release of amphiphilic substances (FFA, lysophosphatidylcholine, etc.), which is conducive to emulsification (Rivera et al., 2018). Under alkaline conditions, hydrolase activity was enhanced, anaerobic hydrolysis was significantly improved, microalgae cell wall was degraded, and long chain fatty acids (LCFA) were accumulated (Qiu et al., 2020).

Oil Extraction

The most usual and simplest method to extract microalgae oil from broken cells is organic solvent extraction. At present, hexane and ethanol have been widely used in extracting edible oil, but chloroform, methane, benzene and other organic solvents are toxic and therefore not applicable. According to the similarity compatibility principle, non-polar solvents dissolve and destroy non-polar lipids on microalgae cell membrane to extract oil. There are not only monophasic solvents (mixtures of one or more polar/non-polar solvents and water miscible, e.g., hexane/ethanol), but also biphasic solvents (mixtures of two or more insoluble substances of different polarity, e.g., hexane in water) effective for the extraction of lipids (Liu et al., 2013; Yap et al., 2014). In a two-phase solvent, the two phases play their respective roles. Non-polar organic solvents destroy the hydrophobic interaction between non-polar/neutral lipids. Polar organic solvents destroy polar lipids and the electrostatic forces and hydrogen bonds between lipids and proteins.

For the reason that organic solvents are toxic, volatile, and difficult to recycle, some green solvents are also used, such as bio-based solvents, ionic liquids, convertible solvents, supercritical fluids, subcritical water, and pressurized solvents. The most extensively used technology is supercritical fluid extraction (SFE). Supercritical fluid is a fluid whose temperature and pressure are higher than its critical point. The most commonly used supercritical fluid is CO_2, which is in gas phase under atmospheric conditions, so the solvent can be largely removed to form a solvent-free extract (Molino et al., 2020). CO_2 is a non-polar solvent to extract non-polar compounds, so substances with opposite polarity, such as water, methanol, ethanol and other polar solvents, can be added as co-solvents to enhance the extraction effect (Albarelli et al., 2018; Patil et al., 2018). Typical supercritical CO_2 (SC-CO_2) extraction process is the compression and transmission of liquid CO_2 into the feed

pump, then heat the oven and push CO_2 in supercritical state into the lipid extraction reaction kettle in the oven, where is also a metering valve to decompression in SC-CO_2. Once the decompression completes, CO_2 will escape from the oven to the environment in the form of gas, and the extracted lipids are collected in a special container (Chen and Walker, 2012). SCF is non-toxic, easy to recycle, and runs at low temperatures, but because of the high price of equipment and production, it current main application is in the production of biodiesel. Now it have accomplished that using dissolved in oil microalgae feed directly in the supercritical fluid to enzymatic interesterification reactor to produce biodiesel, without other expensive method of pumping equipment (Reddy et al., 2014; Drira et al., 2016; Patil et al., 2017; Taher et al., 2020). SCF is an effective alternative method for extracting natural bioactive ingredients (such as astaxanthin, carotenoids, etc.) in the food industry, and has been applied to decaffeination of coffee and extraction of hops, etc. (Krichnavaruk et al., 2008; Goto et al., 2015; Kwan et al., 2018; Rammuni et al., 2019). SCF is conducive to the extraction of unsaturated fatty acids. The unsaturated fatty acids in SC-CO_2 extract are about 40%, while the traditional solvent extract is only 10–15% (Srivastava et al., 2018; de Melo et al., 2020). However, in the production of edible oil, economic and feasible strategies should be formulated according to the cost.

Hydroenzymatic method is a new type of gentle, low energy consumption, highly specific method that does not produce harmful or volatile organic compounds but produce many bioactive substances, which has attracted the attention of scholars in recent years (Sekhon et al., 2008; Gai et al., 2013; Liu et al., 2016; Nadar et al., 2018). Although, direct enzymatic cell degradation is not only costly but also inefficient, so the pretreatment methods mentioned above are often needed before hydroenzymatic oil extraction, such as ultrasound-assisted hydroenzymatic method, which has achieved good results in oil extraction (Hou et al., 2018). The water enzymatic oil extraction technology also has a problem, that is, the emulsifying layer will be formed in the process. In order to improve the oil extraction rate of the water enzymatic method, it is necessary to study the composition and properties of the emulsifying layer and choose appropriate methods for demulsification (Yusoff et al., 2015). Currently, demulsification methods used in the research include centrifugation, phase transformation, freezing and thawing, shearing, microfiltration and other physical and chemical methods (Ahmad et al., 2011a; Zolfaghari et al., 2016; Cai et al., 2019). Wu et al. (2009) used PH regulation and enzyme treatment to provide an effective strategy to destroy the stability of oil-rich emulsions formed during water extraction. Qian et al. (2019) used petroleum ether to eliminate the emulsification in water enzymatic extraction of soybean oil and enriched nearly 99% of the oil in the emulsion. Protein is a natural emulsifier and stabilizer, which can stabilize the emulsion at the oil/water interface. Protein–lipid interaction in the emulsion is closely related to the stability of the emulsion, and relevant mechanism research is still underway. Zhang and Lu (2015) studied the hydroenzymatic extraction of proteins from peanut oil emulsions, revealing the unique structural characteristics of peanut proteins adsorbed on the oil/water interface, as well as the structure-function relationship between enzymatic hydrolysis of proteins and their emulsifying properties. In the water enzymatic extraction of microalgae edible oil, the composition, spatial structure and chemical properties of the emulsion still need to be further studied to reveal the enzymatic hydrolysis and demulsification mechanism, further improve the oil extraction rate and reduce the production cost. Besides, there are other mild oil extraction methods in recent years, such as the three-phase distribution method (TPP) (Panadare and Rathod, 2017). **Table 3** Enamala et al. (2018), Baskar et al. (2019), Karmakar and Halder (2019), Li P. et al. (2019), and Nagappan et al. (2019) shows the comparison of various methods for extracting microalgae oil.

CONCLUSION AND OUTLOOK

Firstly, the exploitation and utilization of microalgae in edible oil is mainly based on the nutritional value of microalgae. By comparing the fat content of some traditional oil materials and oil-producing algae strains above, and taking into account the lipid productivity of microalgae, we believe that microalgae is a sustainable and rich source of oil. In terms of fatty acid composition, as a result of the great difference of fatty acid composition in the cells of each algae species, some rich in ω-3 PUFAs and with reasonable composition of them can be selected for research and development of new food. To comprehensively evaluate the nutritional quality of microalgae, in addition to studying more biochemical components, it is also necessary to conduct *in vitro* simulated digestion experiments to test the digestibility of microalgae oil and evaluate its bioavailability (Niccolai et al., 2019). Currently, microalgae oil, particularly algal oil DHA and EPA, is mainly used as a nutritional fortifier for baby milk powder or health products, and is also added to candies, pasta and beverages (Wang et al., 2015; Rizwan et al., 2018). There are two applications of microalgae oil in edible oil: first, as the raw material of ordinary edible oil. The oil content and fatty acid composition of microalgae oil meet the raw material requirements of ordinary edible oil; Second, as a source of nutritional fortifiers in blending oils. The fatty acid composition of certain algal strains rich in ω-3 PUFAs should be studied in depth, and the processing methods of algal strains should be improved to purify and isolate physiological PUFAs such DHA and EPA as edible oil additives.

Additionally, we consider that the lipid synthesis pathway of microalgae cells is similar to but different from that of higher plants, and it has a unique PUFAs prolongation process. In present paper, the methods of promoting lipid accumulation, cell fragmentation and oil extraction in microalgae cells were introduced from the technical level. It is need to be considered the economic feasibility and commercial prospect of using microalgae as an alternative to conventional plant oil crops. Oleaginous microalgae with fast growth, high oil content, but the production cost is higher, and in the downstream processing, the costs of breaking the cell wall and extracting oil have increased than traditional plant source fuel. And we need improve the microalgae oil production technology from microalgae cultivation, cell broken, oil extraction, and oil processing

TABLE 3 | Comparison of different lipid extraction methods of microalgae (Enamala et al., 2018; Baskar et al., 2019; Karmakar and Halder, 2019; Li P. et al., 2019; Nagappan et al., 2019).

Method	Principle	Example	Advantage	Disadvantage
Organic solvent	Similar phase solution principle.	Hexane, chloroform, methanol, ethyl ether, acetone, et al.	Simple, cheap, convenient for continuous operation, suitable for different production scale.	Solvent toxic, volatile, residual, difficult to separate.
Bio-base solvent	Solvents derived from biological resources.	Terpene such as D-limonene, corydaline and α-alkene and ethyl acetate, 2-methyltetrahydrofuran, ethyl lactate, cyclopentyl methyl ether, et al.	Biodegradable and non-toxic.	Consistent feedstock supply and the thermodynamics and kinetics of large-scale extraction are not well studied.
Ionic liquids	Non-aqueous salt solution consisting of organic cations and anions to extract oil, and the effect of extraction is closely related to the ion structure.	[Bmim][Cl], [Bmim][BF_4], [Bmim][$MeSO_4$], [Bmim][HSO_4], [Bmim][CF_3SO_3], [Emim][Ac], [Emim][$MeSO_4$], [Propy-mim][Br], [Cyno-mim][Br], et al.	Low toxicity, non-volatile property, thermal stability, non-flammable, and liquid at a wide temperature range (0–140°C), can be adjusted for specific solubility, polarity, conductivity, and relative hydrophobicity.	Lack of detailed knowledge and the commercial viability of large-scale applications now.
Supercritical fluids	The solvent is in supercritical fluid state to extract oil.	CO_2, ethanol, glycerin, H_2O, et al.	Low toxicity, solvent free extract produced in the process, can retain the natural quality of bioactive compounds, save energy and reduce environmental pollution.	High infrastructure and operating costs.
Switchable solvent	Polarity, hydrophilicity, or ionic strength can be converted under trigger action.	*N*,*N*-dimethylcyclohexylamine, *N*-ethylbutylamine, dipropylamine, midines, secondary amines, tertiary amines, et al.	Non-toxic/low toxic, low energy, environmentally friendly, high selectivity of required compounds (lipids), and reduced extraction time.	The feasibility of expanding production beyond the laboratory scale needs to be studied.
Subcritical water extraction	The oil was extracted with subcritical water as solvent.	Water.	As a clean solvent, water is environmentally friendly and easy to recycle.	Harsh conditions, water needs to be disinfected to remove dissolved oxygen water, and high temperature on the equipment with corrosion
Pressurized liquid extraction	Pressure increases the boiling point of organic or water-based solvent, and the temperature increases the extraction rate.	CO_2, ethanol, limonene, et al.	Equipment automation, shorter extraction time, less extraction solvent dosage, and less damage to human and environment because of the process of extraction is airtight.	High voltage equipment is expensive.
Aqueous enzymatic method	On the basis of pretreatment of broken cells, enzymes were used to make the oil easy to be released from the oil, and then water was used as a solvent to separate the oil from water-soluble substances (proteins, carbohydrates, etc.).	Cellulase, pectinase, amylase, protease, et al.	Mild, low energy consumption, environmentally friendly, safe and hygienic.	Long time, high cost of enzyme and emulsification problem
Three phase partitioning	After mixing the powder source with the salt solution for a fixed time, tert-butanol was added slowly; and the solution can be divided into oil phase, water phase and intermediate protein phase	Tert-butanol and ammonium sulfate.	Mild, economical and can be directly used in crude plant materials.	It is influenced by different factors (such as pH value, temperature, incubation time, etc.), which need to be further studied

to promote industrialization and application promotion of microalgae oil (Wase et al., 2018; Wang et al., 2019). A modern way to process microalgae is to build microalgae biorefineries, microalgae produce biodiesel and synthesis of a large number of high value compounds, such as pigment, vitamins, PUFAs, antioxidant and so on. Making full use of microalgae resources production is useful to meet energy, chemical, food, medicine, cosmetics, and other products in a wide range of industries. To reduce microalgae processing costs and achieve integrated microalgae product production, a comprehensive economic

and environmental study on the production of high-value compounds from microalgae is required to develop a more economically feasible biorefining route (Lee and Chang, 2017; Mobin et al., 2019; Banu et al., 2020).

What's more, the development of microalgae edible oil is different from biodiesel in that it is necessary to consider the edible safety of the oils, the chemical and physical properties of the products are very important, and toxicological studies of microalgae oils must be conducted to check for potential toxicity (including mutagenicity, systemic toxicity, reproductive and multigenerational toxicity), the possibility of risk levels of heavy metals and pathogenic microorganisms, and the safety of the oil production process (Draaisma et al., 2013). Several DHA rich algal species have been accepted as Generally Recognized as Safe (GRAS) by Food and Drug Administration (FDA), for an instance, *Schizochytrium* sp., *Chlorella protothecoides*, *Ulkenia* sp. *SAM2179* et al. (Szabo et al., 2014). There have been several studies on the toxicological effects of microalgae oil (Kroes et al., 2003; Kagan and Matulka, 2015), but more evidence of safety is needed before microalgae oil can be widely used.

Last but not least, a common problem faced by edible oils is how to improve the oxidative stability and shelf life of algal oils. In particular, microalgae oil contain more PUFAs than normal oils, and thus have less antioxidant capacity. At present, the commonly used antioxidant means are adding natural or synthetic antioxidants to the oil, and the combined oxidants have better effect than the single oxidant, so further research should be conducted to optimize the formulation and mixture ratio of antioxidant combinations (Laguerre et al., 2007; Jacobsen et al., 2008; Gaffney et al., 2014; Shen et al., 2020). Ganiari et al. (2017) used edible and active films and coatings as carriers of natural antioxidants in lipid food, which provid a barrier for water, oxygen and solute movement in food and achieve good antioxidant effect. The latest research applied some nanotechnology to microencapsulate oil or make microemulsions, which not only plays a significant role in preventing oil oxidation, but also can improve the physical and nutritional properties of oil (Ziani et al., 2012; da Silva Santos et al., 2019; Sharma et al., 2019; Linke et al., 2020). Yet, as a new technical method, its production method, performance characterization, safety evaluation and many other aspects need to carry out extensive research.

AUTHOR CONTRIBUTIONS

ZX and WY generally guided the topic selection and research content of the manuscript. YY undertook the main writing work of the manuscript. XG and YZ provided specific guidance and detailed revisions. All authors contributed to the article and approved the submitted version.

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SUPPLEMENTARY MATERIAL

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