Aeroterrestrial and Extremophilic Microalgae as Sources in Cosmetics

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Contributor: Blagoy Uzunov

Microscopic prokaryotic and eukaryotic algae (microalgae), which can be effectively grown in mass cultures, are gaining increasing interest in cosmetics. Up to now, the main attention was on aquatic algae, while species from aeroterrestrial and extreme environments remained underestimated. In these habitats, algae accumulate high amounts of some chemical substances or develop specific compounds, which cause them to thrive in inimical conditions. Among such biologically active molecules is a large family of lipids, which are significant constituents in living organisms and valuable ingredients in cosmetic formulations. Therefore, natural sources of lipids are increasingly in demand in the modern cosmetic industry and its innovative technologies. Among novelties in skin care products is the use of lipid nanoparticles as carriers of dermatologically active ingredients, which enhance their penetration and release in the skin strata.

Keywords: carotenoids ; Cyanoprokaryota ; fatty acids ; morphological type ; PUFA ; Ochrophyta ; Rhodophyta ; Chlorophyta ; Streptophyta ; sterols

1. Introduction

The increasing use of microalgae in the cosmetic industry as extraordinary rich source of novel high-value functional products, obtained in eco-friendly and cost-effective processes, is widely recognized [1][2]. To date, more than 15,000 novel compounds of algal origin have been identified [3]. Many bio-based microalgal products are often “multipurposed” and are applied in dermal cosmetics as sunscreens, skin sensitizers and colorants, as well as agents for moisturizing, water-binding, texturizing, thickening, tanning, whitening, etc. [2][4][5][6][7][8]. In such skin-related applications, the chemically and functionally diverse group of lipids and their derivates comprise a significant gradient [9][10]. Apart from the fact that deficiencies in cutaneous lipids cause discomfort, which may lead to serious skin diseases (e.g., atopic dermatitis, psoriasis, acne, rosacea, hereditary ichthyoses, allergic and irritant contact dermatitis and hidradenitis suppurativa), the broad usage of lipids is based also on their ability to form a protective multifunction skin barrier and on their role as emollients or emulsifiers in the bulk of care and make-up products as well [9][11]. Today, with the flourishing development of nanotechnology, which leads to a fast product innovation [12][13], the potential of lipid nanoparticles (LNP), which seem to be both effective and economic, has been recognized as promising [14]. For example, in dermal cosmetics, about a decade ago, it was already demonstrated that nano-sized sunscreen products have better performance than micron-sized materials [15].

Therefore, currently, nano-sized systems are being increasingly studied and applied to encapsulate active ingredients in order to enhance the efficacy of their percutaneous delivery to targeted cells and are also used to improve the physiochemical stability of skin-based cosmetic products [16]. Such loaded LNP have the advantage of a cumulative effect achieved by a combination of their easier penetration with the enhanced and prolonged release of the carried ingredient to the targeted cellular and subcellular regions [17][18][19]. In this way, the overall functionality of the final product is improved, allowing LNP-based cosmetic formulations to be highly effective in skin protection, in treating dermatological disorders and in antiaging therapy [17][18][19]. In addition, many loaded LNP may provide glowing skin [19]. All these positive effects can be achieved not only by topical skin administrations but also by oral applications [19], and it is easily explainable that the modern dermal cosmetic industry is searching for new high-value functional lipid products of natural origins.

Moreover, in the recent conditions of our society, with growing consumer demands for vegetal oils, there is a rapidly increasing general interest in lipid-rich microalgae (>20% of lipids on a cell dry weight basis), named oleaginous [10][20]. Evidence has been accumulated that the average lipid content in microalgae varies between 1 and 70%, but in certain conditions, it can reach 90% of dry weight (for details, see Reference [21]). This commercial interest is strongly supported by the fact that many oleaginous microalgae can be cultivated at a production scale both autotrophically and heterotrophically, mostly with inexpensive nutrient regimes, and have faster growth rates with high biomass productivity as

compared to terrestrial crops [2][22][23][24]. Further comparisons show that microalgae have even more advantages, since they can be grown all-year-round without the use of arable land, have low water consumption and have low environmental impact [25]. Another important fact that has to be taken into account is the great general biodiversity of microalgae, which is far away from being thoroughly studied and is gaining increasing attention due to recent surge in searching for indigenous commercially important strains or phycoprospecting [2][26][27].

Lipids produced by microalgae belong to two major groups of polar lipids and nonpolar, or neutral lipids [25][28][29]. Polar lipids have an important role in cell structure and cell signaling processes and are commonly known also as structural or membrane lipids [28][29]. They comprise a small part of the total lipid fraction in cells (ca. 20%) and usually have long chains of extractable fatty acids (FtAs) [25][28][29][30]. Nonpolar lipids have diverse biological functions, but most of them, triglycerides in particular, are often pointed out as responsible for energy storage [28]. Most oleaginous microalgae have the capacity to produce significant amounts of nonpolar lipids (up to 80% of the total cell lipid content), the accumulation of which can be influenced, especially in conditions of a lack of nutrients or in stress environments [25][28].

The capacity of microalgae to produce lipids in such appreciable amounts, combined with the advantages of their growing, has stimulated considerable interest in their screening for useful and unusual lipid compositions and their mass cultivation as a feedstock for various biotechnological products [29]. Until now, most research has been oriented towards freshwater, marine and hyperhaline aquatic species from the genera *Aphanizomenon*, *Arthrospira*, *Chlorella*, *Desmodesmus*, *Dunaliella*, *Haematococcus*, *Nannochloropsis*, *Scenedesmus* and *Spirulina* [2]. However, the commercial potential of microalgae inhabiting many other aeroterrestrial or extreme habitats remains untapped [2]. The species from these inimical environments had to develop ultrastructural, physiological and biochemical adaptive features, which include a series of protective natural compounds of special interest for future applications in human life and cosmetics in particular [2]. Moreover, such microalgae can be successfully cultivated in outdoor conditions detrimental for standard crops and other algae [31].

2. Current Insights

The conducted analysis of publications on lipids from AEM issued in the last 56 years showed that most studies addressed PR and FA, specifically carotenoids and FtAs (mainly PUFA), but even these two lipid categories and their accumulation in AEM deserve more attention. In fact, from the eight major groups accepted in LIPID MAPS classification [32][33], only five (i.e., FA, GL, GP, ST, PR and PK) have been examined in different phyla of AEM, as follows: Cyanoprokaryota (FA, GL, ST and PR); Rhodophyta (FA, GL, ST, PR and PK); Ochrophyta (FA, GL and PR); Chlorophyta (FA, GL, GP, ST, PR and PK) and Streptophyta (PK)—**Table 1**. However, according to the data obtained, it is possible to state that stronger efforts are necessary to improve our knowledge regarding GL, GP and SL and their spread in AEM. Special attention has to be paid to SP, which, according to our best knowledge, remain unknown in AEM.

Table 1. Main lipid classes investigated in aeroterrestrial (AET) and extremophilic (EXT) microalgae of different taxonomic phyla and classes. Abbreviations: AA—arachidonic acid, ASX—astaxanthin, DGD—digalactosylglycerol, DGTA—diacylglyceryl-hydroxymethyl-*N*,*N*,*N*-trimethyl-β-Ala, DGTS—diacylglyceryl-*N*,*N*,*N*-trimethylhomo-Ser, EPA—eicosapentaenoic acid, FA—fatty acyls, FtAs—fatty acids, FAEs—fatty acid esters, GL—glycerolipids, GP—glycerophospholipids, MGD—monogalactosylglycerol, PC—phosphatidylcholine/lecithin, PE-phosphatidyletanolamine/cephalin, PG—phosphatidylglycerol, PI—phosphatidylinositol, PK—polyketides, PR—prenol lipids, PS—phosphatidylserine, PSD—phosphatidylglycerol, SQD—sulphoquinovosyl diacylglycerol, ST—sterol lipids, TGD—triagalactosylglycerol. In Bold are outlined AEM, noted in the cited literature as promising for commercial production and relevant lipids.

Taxonomic Group/Alga	Ecological Group	Investigated Lipid Classes with Examples of Detected Lipids	References
CYANOPROKARYOTA			
Anabaena cylindrica	**AET**	**FA (PUFA—linoleic and linolenic acids, SAFA—palmitic acid and MUFA); GL; ST**	**[34]**
***Anabaena cylindrica* 1403-2**	**AET**	**GL (MGD, DGD, SQD and PSD)**	**[35]**
Anabaena vaginicola	**AET**	**PR (lycopene, lutein, beta-carotene, zeaxanthin)**	**[36]**
***Calothrix* sp.**	**AET**	**ST**	**[34][37][38][39][40]**

Taxonomic Group/Alga	Ecological Group	Investigated Lipid Classes with Examples of Detected Lipids	References
Desmonostoc muscorum	AET	FA (PUFA—hexadecadienoic and linoleic acids, SAFA—palmitic acid, MUFA—oleic and palmitoleic acids); GL; ST	[34][37][38][39][40]
Drouetiella lurida	AET—soil, subaerial	ST (seven unsaturated ST)	[41]
Microcoleus autumnalis	AET—soil, subaerial	ST (cholesterol, β-sitosterol and stigmasterol with squalene as a precursor; ergosterol)	[40]
Nostoc calcarea	AET—soil, subaerial	PR (lycopene, lutein, beta-carotene, zeaxanthin)	[36]
Nostoc calcicola B 1459-2	AET—soil, subaerial	FA (PUFA—linolenic acid, SAFA, MUFA), GL—MGD, DGD, SQD and PSD	[35]
Nostoc carneum	AET—soil, subaerial	ST	[34][37][38][39][40]
"Nostoc canina"	AET (symbiont?)	FA (PUFA— linoleic acid, SAFA -palmitic acid, MUFA—palmitoleic and oleic acids); GL; ST (cholesterol and lanosterol)	[34]
Nostoc commune	AET	ST	[34][37][38][39][40]
Nostoc commune var. *sphaeroides*	AET	ST (campesterol, sitosterol and clionasterol)	[42][39][43]
Nostoc punctiforme PCC73102	AET	FA (FAEs—oxylipins), PR (genes for ASX and canthaxanthin)	[44][45]
Nostoc sp. PCC7120	AET	FA (FAEs—oxylipins)	[46]
Oscillatoria chalybea B1459-2	AET	GL (MGD, DGD, TGD, SQD and PSD)	
Oscillatoria sp. PBGA3	AET—soil	FA (FtAs)	[47]
Scytonema sp.	AET	ST (cholest-5-en-3β-ol (18.9 %), 3β-methoxycholest-5-ene (16.2 %) and 3β-acetoxycholest-5-ene (11.2 %), ergosta-5,7,22,24(28)-tetraen-3β-ol)	[48]
Tolypothrix tenuis B1482-3	AET	GL (MGD, DGD, TGD, SQD and PSD)	[35]
Tolypothrix sp. PBGA1	AET	FA (FtAs)	[47]
Tolypothrix sp. PBGA2	AET	FA (FtAs)	[47]
RHODOPHYTA			
Cyanidium caldarium	EXT—thermal springs	GL; ST (ergosta-5,7,22,24(28)-tetraen-3β-ol)	[49]
Cyanidioschyzon merolae	EXT—thermal springs	PK	[50]
Galdieria sulfuraria (>47 strains)	EXT—thermal springs/AET—cryptoendolith	FA (PUFA—linoleic and linolenic acid, SAFA—palmitic acid, MUFA—oleic and palmitoleic acids); GL (MGD, DGD and SQD; PG, PC, PE, PI, PS and phosphatidate); GP; ST (ergosta-5,7,22,24(28)-tetraen-3β-ol and ergosterol); PR (ß-carotene, lutein); PK	[51][52][53][49][50]
Galdieria sulfuraria/Galdieria sp.	EXT—acidic but non-thermophilic	FA (PUFA—linoleic and linolenic acids, SAFA—palmitic, myristic and stearic acids, MUFA—oleic and palmitoleic acids)	[51][52]
Galdieria sp. USB-GBX-832	EXT—thermal springs	FA (PUFA—linoleic acid, AA and EPA; SAFA—palmitic and stearic acid, MUFA—oleic acid)	[54]
Pophyridium purpureum	AET-soil	FA (PUFA—AA and EPA); GL	[55]
OCHROPHYTA Eustigmatophyceae			
Monodopsis subterraneus	AET—soil	FA (PUFA—EPA), GL (DGD)	[56][57][58][59][60][61]

Taxonomic Group/Alga	Ecological Group	Investigated Lipid Classes with Examples of Detected Lipids	References
Monodus guttula	AET	PR (tocopherols)	[2]
Monodus sp.	AET	PR (carotenoids—ASX, beta-carotene and lutein)	[62]
Vischeria/Eustigmatos	AET—soil, subaerial	PR (total carotenoids; ASX, beta-carotene, lutein and canthaxanthin)	[62]
Tribophyceae (=Xanthophyceae)			
Botrydiopsis interdecens	AET	PR (tocopherols)	[2]
Heterococcus sp.	AET	PR (tocopherols)	[2]
Xanthonema sp.	AET	PR (tocopherols)	[2]
CHLOROPHYTA			
Acutodesmus dissociatus TGA1	AET—soil	FA (SAFA—palmitic acid and MUFA—oleic acid)	[47]
Auxenochlorella protothecoides	AET/EXT—acidic	PR (carotenoids—lutein)	[63][64][65][66][67][68][69]
Auxenochlorella pyrenoidosa	AET	PR (carotenoids—ASX, zeaxanthin, canthaxanthin, lutein)	[70][71][72][73]
Bracteacoccus sp.	AET	PR (tocopherols)	[2]
Chlamydocapsa sp.	EXT—snow	PR (canthaxanthin, tocopherols)	[2][74]
Chlamydomonas nivalis	EXT—snow	PR (ASX, canthaxanthin)	[74][75][76]
Chlamydomonas reinhardtii	AET	FA (hydrocarbons—C17 alkene n-heptadecene), GL (betaine lipids—DGTS); GP; ST (ergosterol); PK	[77][78][79][50][80][81]
Chlainomonas sp.	EXT—snow	PR (ASX)	[82]
Chlorella sorokiniana	AET	ST (ergosterol)	[79]
Chlorella variabilis	AET	PK	[50][80][81]
Chlorella variabilis NC64A	AET (symbiotic)	ST (ergosterol)	[79]
Chlorella vulgaris	AET	FA (free FtAs, FAEs—lactones; hydrocarbons—NC64A eptadecane pentadecane, as well as 7- and 8-heptadecene); GL; ST (ergosterol, 7-dehydroporiferasterol, ergosterol peroxide, 7-dehydroporiferasterol per-oxide and 7-oxocholesterol); PR (carotenoids—ASX, zeaxanthin, canthaxanthin and lutein), PK	[21][24][83][84][77][85][70][71][72][73]
Chlorella sp. PGA2	AET—soil	FA (SAFA, MUFA)	[47]
Chlorella sp. TGA2	AET—soil	FA (SAFA- palmitic acid, MUFA—oleic acid)	[47]
Chlorella sp. TGA4	AET—soil	FA (SAFA, MUFA)	[47]
Chlorococcum sp. (1)	AET	PR (carotenoids—ASX (in a free form and as esters), adonixanthin (in a free form and as esters), lutein, canthaxanthin and β-carotene)	[86][87][88][89][90][91][92]
Chlorococcum sp. MA-1	AET	PR (total carotenoids; ASX, lutein, canthaxanthin and ß-carotene)	[88]
Chlorococcum spp.	EXT—snow	PR (ß-carotene, lutein and canthaxanthin)	[51][93][74][75][76][94][95]
Chloroidium ellipsoideum	AET	PR (carotenoids—zeaxanthin)	[96]
Chloromonas alpina	EXT—snow	FA (PUFA, SAFA, MUFA), PR (ASX)	[51][93][97][98][75][76][94][95]
Chloromonas hindakii	EXT—snow	FA (PUFA—α-linolenic, stereadonic and hexadecatetraenoic acids, SAFA—palmitic acid and MUFA—oleic acid); GP; PR (ASX)	[51][93][97][98][75][76][94][95]

Taxonomic Group/Alga	Ecological Group	Investigated Lipid Classes with Examples of Detected Lipids	References
Chloromonas nivalis	EXT—snow	FA (PUFA—hexadecatetraenoic, SAFA and MUFA); PR (ASX, canthaxanthin)	[51][93][97][98][74][75][76][94][95]
Chloromonas nivalis subsp. *tatrae*	EXT—snow	FA (PUFA, SAFA and MUFA); PR (ASX)	[98]
Chloromonas polyptera	EXT—snow	FA (PUFA, SAFA and MUFA), PR (ASX)	[51][93][97][98][75][76][94][95]
Chloromonas remiasii CCCryo 005–99	EXT—snow	FA (PUFA-hexadecatetraenoic acid, SAFA and MUFA), PR	[99][51][93][97][98][75][76][94][95]
Chloromonas spp.	EXT—snow	FA (PUFA, SAFA—palmitic acid and MUFA—oleic acid), PR	[93][97][98][75][76][94][95]
Chromochloris zofingiensis	AET	PR (carotenoids—ASX, canthaxanthin, zeaxanthin, lutein and β-carotene)	[70][86][87][100][101][102][103][104][105][106][107][108][109]
Coccomyxa acidophila	EXT—acidic	PR (carotenoids—ß-carotene and lutein)	[110]
Coccomyxa subellipsoidea	AET	ST (phytosterols)	[79]
Coccomyxa subellipsoidea C-169	AET	PK	[50][80][81]
Coccomyxa sp.	AET	PR (tocopherols)	[2]
Coelastrella oocystiformis	AET	PR (carotenoids—ASX esters and canthaxanthin)	[101][109]
Coelastrella striolata var. *multistriolata*	AET—subaerial, soils	FA (PUFA—linoleic acid, SAFA—palmitic acid and MUFA—oleic acid); PR (carotenoids—canthaxanthin, ASX and ß-carotene)	[111][112]
Dunaliella acidophila	EXT-acidic	FA (PUFA—linolenic, γ-linolenic and linoleic acids; SAFA; MUFA—oleic and elaidic acids; FAEs—lactones, methyl (12R)-hydroxyoctadeca-9Z,13E,15Z-trienoate, methyl (9S)-hydroxyoctadeca-10E, 12Z,15Z-trienoate and methyl ricinoleate; triacylglycerols—trilinolenin, triolein, trielaidin and tristearin); ST (β-sitosterol, isofucosterol, 24-methylenlophenol, (24S)-methyllophenol and two unidentified sterols, acylsterols and phytol); PR (lycopene, alpha-, beta and gamma-carotene)	[113][77]
Edaphochlamys debaryana	AET—soil	FA (FAEs—oxylipins)	[114]
Hindakia tetrachotoma PGA1	AET—soil	FA (SAFA—palmitic acid and MUFA—oleic acid)	[47]
Monoraphidum sp.	EXT—ice	FA (PUFA)	[31]
Muriella terrestris	AET	PR (tocopherols)	[2]
Muriellopsis sp.	AET	PR (carotenoids—lutein)	[115][116][117][118][119]
Neochloris wimmeri	AET	PR (carotenoids—ASX esters and canthaxanthin)	[101][109]
Parietochloris alveolaris	AET-—oil, symbiont	FA (PUFA—EPA, AA and its precursor dihomo-γ-linolenic acid)	[120][121]
Parietochloris alveolaris K-1	AET	FA (PUFA—α-linolenic acid and EPA)	[122][123]
Protosiphon botryoides	AET—soil	PR (carotenoids—ASX esters and canthaxanthin)	[101][109]
Pseudochoricystis ellipsoidea MBIC11204	EXT—thermal springs	FA (FtAs and FAEs—hydrocarbons and triacylglycerols)	[124]
Raphidonema sempervirens	EXT—snow	FA (PUFA, SAFA and MUFA); PR (ß-carotene, ASX, lutein and tocopherols)	[2][93][97][98][74]
Sanguina aurantia	EXT—snow	PR (ASX)	[125]

Taxonomic Group/Alga	Ecological Group	Investigated Lipid Classes with Examples of Detected Lipids	References
Sanguina nivalis	**EXT—snow**	**PR (ASX)**	[125]
Scenedesmus vacuolatus	**AET**	**PR (carotenoids—ASX esters and canthaxanthin)**	[101][109]
***Scenedesmus* spp.**	**AET**	**PR (total carotenoids, ASX and lutein)**	[112][126][127]
Stichococcus bacillaris	**AET**	**PR (tocopherols)**	[2]
***Tetracystis* sp.**	**AET/EXT—cryotolerant**	**PR (canthaxanthin)**	[128]
Tetradesmus obliquus	**AET**	**FA (PUFA—linolenic, linoleic and linolelaidic acids and SAFA—oleic acid); PR (carotenoids—ASX and lutein)**	[21][25][112][129] [130][131][132] [133][126][127]
***Tetradesmus obliquus* (strain *Scenedesmus obliquus* SNW-N)**	**AET**	**PR (lutein)**	[126]
***Tetradesmus obliquus* (strain *Scenedesmus obliquus* FSP-3)**	**AET**	**PR (lutein)**	[134]
Ulothrix zonata	**EXT—ice**	**FA (PUFA)**	[111]
“Unidentified Chlamydomonadaceae”	**EXT—snow**	**FA (PUFA, SAFA and MUFA); PR (ASX)**	[47]
Unidentified “Chlamydomonadales species” TGA3	**AET—soil, thermotolerant**	**FA (SAFA and MUFA)**	[47]
Unidentified “Chlamydomonadales species” TGA5	**AET—soil**	**FA (SAFA and MUFA)**	[47]
STREPTOPHYTA			
Klebsormidium flaccidum	**AET**	**PK**	[50]

The taxonomic diversity of the examined AEM, expressed by the number of species, is as follows: Cyanoprokaryota—20, Rhodophyta—6, Ochrophyta—7, Chlorophyta—53 and Streptophyta—1 (**Figure 1**A and **Table 1**). Thus, AEM from green evolutionary lineages (Chlorophyta and Streptophyta) are the most investigated, but a strong discrepancy between the studies inside the green lineages is obvious, with only one species from Streptophyta (i.e., *Klebsormidium flaccidum*) investigated in comparison with 53 species of 34 genera from Chlorophyta. The next most-studied group is represented by prokaryotic blue–green algae, Cyanoprokaryota, from which 20 species of nine genera have been analyzed. The number of species investigated in Rhodophyta and Ochrophyta is significantly lower and almost similar (six and seven, respectively), but more genera have been covered by studies on Ochrophyta (seven) than from Rhodophyta (four). However, much more strains from Rhodophyta have been examined, with more than 47 strains investigated from a single species (i.e., the extremophilic *Galdieria sulfuraria*). Most of the studied AEM are unicellular (59), followed by filamentous (26) and coenobial algae (2). The highest morphological diversity has been found in the studied chlorophyte algae, while, from Cyanoprokaryota, only filamentous species and, from Rhodophyta, only unicellular species were examined for different lipids.

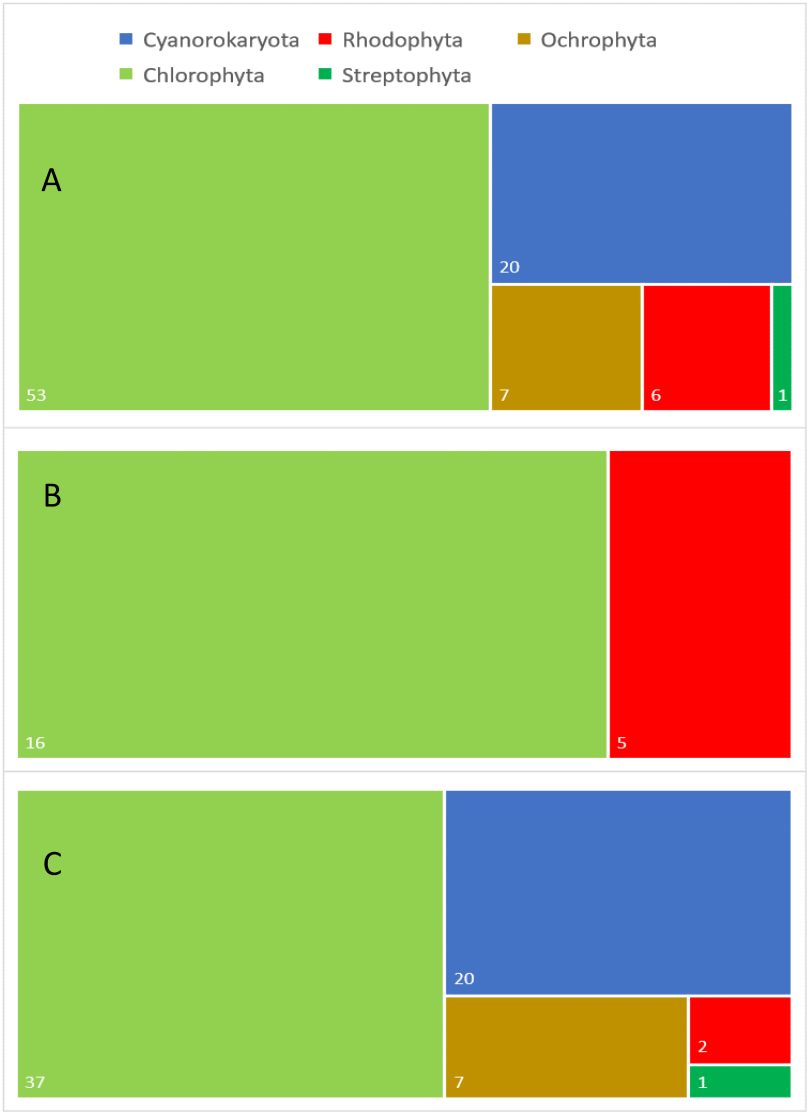

Figure 1. Taxonomic diversity of aeroterrestrial and extremophilic microalgae analyzed for different lipids: (**A**) general taxonomic diversity of the analyzed algal species, (**B**) taxonomic diversity of the examined extremophilic species and (**C**) taxonomic diversity of the investigated aeroterrestrial species. Numbers in white indicate the exact number in each category.

Regarding the ecological affiliation of the examined microalgae (**Figure 1**B,C), it could be stated that the highest number (66) of studied species are aeroterrestrial, and they are the most taxonomically diverse, including representatives from all five major algal phyla (**Figure 1**C). By contrast, the number of examined extremophilic species is lower (21), with only two phyla studied (i.e., Rhodophyta and Chlorophyta) (**Figure 1**B). Despite this generally low number, ten extremophilic algae (ca. 50%) have been considered as promising lipid sources (**Table 1**). However, it has to be boldly underlined that this analysis is based only on Latin species names provided in the available publications, since, for most of the strains, there are neither data from genetic studies nor detailed morphological and ecological descriptions. In the recent times of rapid taxonomical changes, providing such data in future publications is strongly recommended in order to obtain more precise information on the biochemical compositions of the studied microalgae.

The results obtained clearly showed that very few known AEM have been investigated for their lipid content, with most of the studies being quite scarce and oriented towards certain compounds. Less studies were comparative, but the differences in lipid contents were demonstrated with field materials and cultivated in different conditions algae, or ecologically different strains (e.g., thermophilic and non-thermophilic) were investigated. These results may stimulate further research for the best physiological and cultural conditions, which would lead to the optimal yield of algal biomass and certain lipids. As we have shown in the text above, data on all lipid classes in AEM are far away from being complete, and more investigations on certain compounds and in more AEM are needed in the future. Nevertheless, all collected evidence until now suggests the great potential of AEM as novel commercial lipid sources for versatile cosmetic substances and products for skin care. In this regard AEM are comparable with their aquatic (marine and freshwater) counterparts and land plants, which have been much more intensively studied [1][2][135]. Twenty-three AEM have been already pointed out as promising for obtaining certain compounds, most of them from Chlorophyta (**Table 1**). The potential of AEM as beneficial lipid sources can be recognized in two aspects, separately or in combination: (1) quantitative, since, in some AEM, the contents of valuable lipids are higher in comparison with other algae or plants (e.g., *Parietochloris alveolaris* is considered to be the richest natural source of the high value polyunsaturated ω6 AA, or *Chlorodium*

ellipsoideum, in which zeaxanthin exceeded more than nine times that of red pepper, a plant source of zeaxanthin) and (2) qualitative, since, in some AEM, rare and unusual lipids were discovered (e.g., the three hydroxy FtAs in *Dunaliella acidophila*, such as methyl (12R)-hydroxyoctadeca-9Z,13E,15Z-trienoate, methyl (9S)-hydroxyoctadeca-10E, 12Z,15Z-trienoate and methyl ricinoleate, or the unidentified ST of *Scytonema* sp. and *Dunaliella acidophila*). These compositional peculiarities in AEM, as it has been shown earlier, are due to their specific ecology and adaptations to survive inimically in other organism environments, and this potential is still untapped [2]. However, among the AEM examined for lipids occur species from genera that have already been recognized as potential toxin producers or allergic-causative agents [33][136]. On that account, in order to answer safety concerns, we strongly underline that all algae, chosen as lipid sources for direct use or for transformation to LNP, must be subjected to chemical analyses before introducing them into mass cultures and cosmetics formulations.

3. Conclusions

The cosmetics industry is increasingly exploring new compounds derived from natural products, preferably from plant origins, due to strong consumer demands [2][135]. In addition, nanotechnologies are delving deeper into people's lives, including the field of skin cosmetics [16]. It is well-known that skin lipid compositions and structures are significant for proper skin functioning, and their deficit leads to skin diseases and disorders [9][10][11]. Therefore, in addition to traditional topical and oral lipid applications, there is a rising interest in enhancing the targeted lipid transport to different skin layers using nanosized systems, including LNP [12][13][14][15][16][17][18][19]. Currently, different methods for obtaining nanoparticles from lipids have been developed and standardized [14][19]. Since algae from different ecological and taxonomic groups are rich in biologically active compounds, including lipids, they are increasingly becoming the focus of biotechnology [1][2][3][4][5][6][7][8]. However, to date, mostly aquatic micro- and macroalgae have been investigated, whereas the algae from aeroterrestrial and versatile extremophilic habitats have been more neglected. At the same time, due to their peculiar ecology, AEM produce many specific compounds that are not available from other algae or accumulate higher amounts of other chemicals than other algae and plants [2]. To the best of our knowledge, there are no commercially produced skin care preparations, or LNP, yet based on lipids from AEM, despite some AEM being outlined as promising commercial lipid sources (**Table 1**). Therefore, we summarized the available, but quite scattered, data on the lipid contents of different species of AEM issued during the last 56 years.

According to the analysis of this knowledge, considering: (1) the successes in the clinical experiments for the treatment, alleviation or prevention of different skin disorders, as well as all other beneficial effects of versatile lipids, which occur in AEM; (2) the fact that both standard and unusual lipids have been detected, and most of them can be obtained in profitable amounts from AEM; (3) the possibilities to benefit from the direct application of lipids or from their enhanced penetration using LNP; (4) the developed methods for obtaining LNP [13]; (5) the advantages of the effective mass cultivation of AEM species even outdoor, unfavorable for algae from "standard" aquatic habitats, conditions, which can make more cost-effective and beneficial yields of both unusual and standard lipids; (6) the gradually increasing phycoprospection in the background of the enormous biodiversity of microalgae, which has been recognized but is far away from being utilized, we believe that all the provided data will serve as the groundwork to enhance and further encourage studies for broader applications of AEM-derived lipids in novel products of the future dermal cosmetics bioindustry.

References

1. Ariede, M.B.; Candido, T.M.; Jacome, A.L.M.; Velasco, M.V.R.; de Carvalho, J.C.M.; Baby, A.R. Cosmetic attributes of algae—A review. Algal Res. 2017, 25, 483–487.
2. Stoyneva-Gärtner, M.; Uzunov, B.; Gärtner, G. Enigmatic microalgae from aeroterrestrial and extreme habitats in cosmetics: The potential of the untapped natural sources. Cosmetics 2020, 7, 27.
3. Sun, Z.; Li, T.; Zhou, Z.; Jiang, Y. Microalgae as a source of lutein: Chemistry, biosynthesis, and carotenogenesis. In Microalgae Biotechnology. Advances in Biochemical Engineering/Biotechnology; Posten, C., Feng, C.S., Eds.; Springer: Cham, Switzerland, 2016; Volume 153, pp. 37–58.
4. Joshi, S.; Kumari, R.; Upasani, V.N. Applications of algae in cosmetics: An overview. Int. J. Innov. Res. Sci. Eng. Technol. 2018, 7, 1269–1278.
5. Jahan, A.; Ahmad, I.Z.; Fatima, N.; Ansari, V.A.; Akhtar, J. Algal bioactive compounds in the cosmeceutical industry: A review. Phycologia 2017, 56, 410–422.

6. Wang, H.M.D.; Chen, C.C.; Huynh, P.; Chang, J.S. Exploring the potential of using algae in cosmetics. Bioresour. Technol. 2015, 184, 355–362.
7. Basily, H.S.; Nassar, M.M.; Diwani, G.I.; Abo El-Enin, S.A. Exploration of using the algal bioactive compounds for cosmeceuticals and pharmaceutical applications. Egypt. Pharm. J. 2018, 17, 109–120. Available online: http://www.epj.eg.net/text.asp?2018/17/2/109/240673 (accessed on 25 November 2021).
8. Rastogi, R.P.; Sinha, R.P. Biotechnological and industrial significance of cyanobacterial secondary metabolites. Biotechnol. Adv. 2009, 27, 521–539.
9. Bonnet, C. Lipids, a natural raw material at the heart of cosmetics innovation. OCL 2018, 25, D501.
10. De Luca, M.; Pappalardo, I.; Limongi, A.R.; Viviano, E.; Radice, R.P.; Todisco, S.; Martelli, G.; Infantino, V.; Vassallo, A. Lipids from microalgae for cosmetic applications. Cosmetics 2021, 8, 52.
11. Knox, S.; O'Boyle, N.M. Skin lipids in health and disease: A review. Chem. Phys. Lipids 2021, 236, 105055.
12. Kabri, T.; Arab-Tehrany, E.; Belhaj, N.; Linder, M. Physico-chemical characterization of nano-emulsions in cosmetic matrix enriched on omega-3. J. Nanobiotechnol. 2011, 9, 41.
13. Sarkar, R.D.; Singh, H.B.; Kalita, M.C. Enhanced lipid accumulation in microalgae through nanoparticle-mediated approach, for biodiesel production: A mini-review. Heliyon 2021, 7, e08057.
14. Khater, D.; Nsairat, H.; Odeh, F.; Saleh, M.; Jaber, A.; Alshaer, W.; Al Bawab, A.; Mubarak, M.S. Design, preparation, and characterization of effective dermal and transdermal lipid nanoparticles: A review. Cosmetics 2021, 8, 39.
15. Smijs, T.G.; Pavel, S. Titanium dioxide and zinc oxide nanoparticles in sunscreens: Focus on their safety and effectiveness. Nanotechnol. Sci. Appl. 2011, 4, 95.
16. Khezri, K.; Saeedi, M.; Dizaj, S.M. Application of nanoparticles in percutaneous delivery of active ingredients in cosmetic preparations. Biomed. Pharmacother. 2018, 106, 1499–1505.
17. Alvarez, A.M.R.; Rodríguez, M.L.G. Lipids in pharmaceutical and cosmetic preparations. Grasas Aceites 2000, 51, 74–96. Available online: https://grasasyaceites.revistas.csic.es/index.php/grasasyaceites/article/view/409 (accessed on 3 November 2021).
18. Zielinska, A.; Nowak, I. Fatty acids in vegetable oils and their importance in cosmetic industry. CHEMIK Nauka Tech. Rynek 2014, 68, 103–110.
19. Ahmad, J. Lipid nanoparticles based cosmetics with potential application in alleviating skin disorders. Cosmetics 2021, 8, 84.
20. Patel, A.; Karageorgou, D.; Rova, E.; Katapodis, P.; Rova, U.; Christakopoulos, P.; Matsakas, L. An overview of potential oleaginous microorganisms and their role in biodiesel and omega-3 fatty acid-based industries. Microorganisms 2020, 8, 434.
21. Mata, T.M.; Martins, A.A.; Caetano, N.S. Microalgae for biodiesel production and other applications: A review. Renew. Sustain. Energy Rev. 2010, 14, 217–232.
22. Spolaore, P.; Joannis-Cassan, C.; Duran, E.; Isambert, A. Commercial applications of microalgae. J. Biosci. Bioeng. 2006, 101, 87–96.
23. McKie-Krisberg, Z.M.; Laurens, L.M.L.; Huang, A.; Polle, J.E.W. Comparative energetics of carbon storage molecules in green algae. Algal Res. 2018, 31, 326–333.
24. Safi, C.; Zebib, B.; Merah, O.; Pontalier, P.-Y.; Vaca-Garcia, C. Morphology, composition, production, processing and applications of Chlorella vulgaris: A review. Renew. Sustain. Energy Rev. 2014, 35, 265–278.
25. Pignolet, O.; Jubeau, S.; Vaca-Garcia, C.; Philippe, M. Highly valuable microalgae: Biochemical and topological aspects. J. Ind. Microbiol. Biotechnol. 2013, 40, 781–796.
26. Wilkie, A.C.; Edmundson, S.J.; Duncan, J.G. Indigenous algae for local bioresource production: Phycoprospecting. Energy Sustain. Dev. 2011, 15, 365–371.
27. Stoyneva-Gärtner, M.; Uzunov, B.; Gärtner, G.; Radkova, M.; Atanassov, I.; Atanassova, R.; Borisova, C.; Draganova, P.; Stoykova, P. Review on the biotechnological and nanotechnological potential of the streptophyte genus Klebsormidium with pilot data on its phycoprospecting and polyphasic identification in Bulgaria. Biotechnol. Biotechnol. Equip. 2019, 33, 559–578.
28. Aratboni, H.A.; Rafiei, N.; Garcia-Granados, R.; Alemzadeh, A.; Morones-Ramírez, J.R. Biomass and lipid induction strategies in microalgae for biofuel production and other applications. Microb. Cell Fact. 2019, 18, 178.
29. Slocombe, S.P.; Zhang, Q.Y.; Ross, M.; Anderson, A.; Thomas, N.J.; Lapresa, Á.; Rad-Menéndez, C.; Campbell, C.N.; Black, K.D.; Stanley, M.S.; et al. Unlocking nature's treasure-chest: Screening for oleaginous algae. Sci. Rep. 2015, 5,

9844.

30. Richter, C.K.; Skulas-Ray, A.C.; Kris-Etherton, P.M. Recommended intake of fish and fish oils worldwide. In Fish and Fish Oil in Health and Disease Prevention; Raatz, S.K., Bibus, D.M., Eds.; Elsevier: London, UK, 2016; pp. 27–48.
31. Řezanka, T.; Nedbalová, L.; Lukavský, J.; Střížek, A.; Sigler, K. Pilot cultivation of the green alga Monoraphidium sp. producing a high content of polyunsaturated fatty acids in a low-temperature environment. Algal Res. 2017, 22, 160–165.
32. Fahy, E.; Subramaniam, S.; Brown, H.A.; Glass, C.K.; Merrill, A.H.; Murphy, R.C.; Raetz, C.R.H.; Russell, D.W.; Seyama, Y.; Shaw, W.; et al. A comprehensive classification system for lipids. J. Lipid Res. 2005, 46, 839–861.
33. Fahy, E.; Subramaniam, S.; Murphy, R.C.; Nishijima, M.; Raetz, C.R.H.; Shimizu, T.; Spener, F.; van Meer, G.; Wakelam, M.J.O.; Dennis, E.A. Update of the LIPID MAPS comprehensive classification system for lipids. J. Lipid Res. 2009, 50, S9–S14.
34. Sallal, A.K.; Nimer, N.A.; Radwan, S.S. Lipid and fatty acid composition of freshwater cyanobacteria. Microbiology 1990, 136, 2043–2048.
35. Zepke, H.D.; Heinz, E.; Radunz, A.; Linscheid, M.; Pesch, R. Combination and positional distribution of fatty acids in lipids from blue-green algae. Arch. Microbiol. 1978, 119, 157–162.
36. Hashtroudi, M.S.; Shariatmadari, Z.; Riahi, H.; Ghassempour, A. Analysis of Anabaena vaginicola and Nostoc calcicola from Northern Iran, as rich sources of major carotenoids. Food Chem. 2013, 136, 1148–1153.
37. Fagundes, M.B.; Wagner, R. Sterols biosynthesis in algae. In Bioactive Compounds. Biosynthesis, Characterization and Applications; Zepka, L.Q., Ed.; IntechOpen: London, UK, 2021.
38. Konlhase, M.; Pohl, P. Saturated and unsaturated sterols of nitrogen-fixing blue-green algae (cyanobacteria). Phytochemistry 1988, 27, 1735–1740.
39. Paoletti, C.; Pushparaj, B.; Florenzano, G.; Capella, P.; Lercker, G. Unsaponifiable matter of green and blue-green algal lipids as a factor of biochemical differentiation of their biomasses: II. Terpenic alcohol and sterol fractions. Lipids 1976, 11, 266–271.
40. Fagundes, M.B.; Falk, R.B.; Facchi, M.M.X.; Vendruscolo, R.G.; Maroneze, M.M.; Zepka, L.Q.; Wagner, R. Insights in cyanobacteria lipidomics: A sterols characterization from Phormidium autumnale biomass in heterotrophic cultivation. Food Res. Int. 2019, 119, 777–784.
41. De Souza, N.J.; Nes, W.R. Sterols: Isolation from a bluegreen alga. Science 1968, 162, 363.
42. Luo, X.; Su, P.; Zhang, W. Advances in microalgae-derived phytosterols for functional food and pharmaceutical applications. Mar. Drugs 2015, 13, 4231–4254.
43. Rasmussen, H.E.; Blobaum, K.R.; Park, Y.-K.; Ehlers, S.J.; Lu, F.; Lee, J.-Y. Lipid extract of Nostoc commune var. sphaeroides Kützing, a blue-green alga, inhibits the activation of sterol regulatory element binding proteins in HepG2 Cells. J. Nutr. 2008, 138, 476–481.
44. Lang, I.; Feussner, I. Oxylipin formation in Nostoc punctiforme (PCC73102). Phytochemistry 2007, 68, 1120–1127.
45. Steiger, S.; Sandmann, G. Cloning of two carotenoid ketolase genes from Nostoc punctiforme for the heterologous production of canthaxanthin and astaxanthin. Biotechnol. Lett. 2004, 26, 813–817.
46. Lang, I.; Göbel, C.; Porzel, A.; Heilmann, I.; Feussner, I. A lipoxygenase with linoleate diol synthase activity from Nostoc sp. PCC 7120. Biochem. J. 2008, 410, 347–357.
47. Thangavel, K.; Krishnan, P.R.; Nagaiah, S.; Kuppusamy, S.; Chinnasamy, S.; Rajadorai, J.S.; Olaganathan, G.N.; Dananjeyan, B. Growth and metabolic characteristics of oleaginous microalgal isolates from Nilgiri biosphere Reserve of India. BMC Microbiol. 2018, 18, 1.
48. Řezanka, T.; Dembitsky, V.M.; Go, J.V. Sterol compositions of the filamentous nitrogen-fixing terrestrial cyanobacterium Scytonema sp. Folia Microbiol. 2003, 48, 357–360.
49. Seckbach, J.; Ikan, R.; Ringelberg, D.; White, D. Sterols and phylogeny of the acidophilic hot springs algae Cyanidium caldarium and Galdieria sulphuraria. Phytochemistry 1993, 34, 1345–1349.
50. Shelest, E.; Heimerl, N.; Fichtner, M.; Sasso, S. Multimodular type I polyketide synthases in algae evolve by module duplications and displacement of AT domains in trans. BMC Genomics 2015, 16, 1015.
51. Procházková, L.; Remias, D.; Řezanka, T.; Nedbalová, L. Ecophysiology of Chloromonas hindakii sp. nov. (Chlorophyceae), causing orange snow blooms at different light conditions. Microorganisms 2019, 7, 434.
52. Graziani, G.; Schiavo, S.; Nicolai, M.A.; Buono, S.; Fogliano, V.; Pinto, G.; Pollio, A. Microalgae as human food: Chemical and nutritional characteristics of the thermo-acidophilic microalga Galdieria sulphuraria. Food Funct. 2013, 4,

144–152.

53. Vítová, M.; Goecke, F.; Sigler, K.; Řezanka, T. Lipidomic analysis of the extremophilic red alga Galdieria sulphuraria in response to changes in pH. Algal Res. 2016, 13, 218–226.
54. López, G.; Yate, C.; Ramos, F.A.; Cala, M.P.; Restrepo, S.; Baena, S. Production of polyunsaturated fatty acids and lipids from autotrophic, mixotrophic and heterotrophic cultivation of Galdieria sp. strain USBA-GBX-832. Sci. Rep. 2019, 9, 10791.
55. Guil-Guerrero, J.; Belarbi, E.H.; Rebolloso-Fuentes, M. Eicosapentaenoic and arachidonic acids purification from the red microalga Porphyridium cruentum. Bioseparation 2000, 9, 299–306.
56. Iwamoto, H.; Sato, S. Production of EPA by freshwater unicellular algae. J. Am. Oil. Chem. Soc. 1968, 71, 434.
57. Cohen, Z. Production potential of eicosapentaenoic acid by Monodus subterraneus. J. Am. Oil Chem. Soc. 1994, 71, 941–945.
58. Khozin-Goldberg, I.; Didi-Cohen, S.; Cohen, Z. Biosynthesis of eicosapentaenoic acid (EPA) in the freshwater eustigmatophyte Monodus subterraneus (Eustigmatophyceae). J. Phycol. 2002, 38, 745–756.
59. Lu, C.; Rao, K.; Hall, D.; Vonshak, A. Production of eicosapentaenoic acid (EPA) in Monodus subterraneus grown in a helical tubular photobioreactor as affected by cell density and light intensity. J. Appl. Phycol. 2001, 13, 517–522.
60. Vazhappilly, R.; Chen, F. Eicosapentaenoic acid and docosahexaenoic acid production potential of microalgae and their heterotrophic growth. J. Am. Oil Chem. Soc. 1998, 75, 393–397.
61. Khozin-Goldberg, I.; Cohen, Z. The effect of phosphate starvation on the lipid and fatty acid composition of the fresh water eustigmatophyte Monodus subterraneus. Phytochemistry 2006, 67, 696–701.
62. Stoyneva-Gärtner, M.; Stoykova, P.; Uzunov, B.; Dincheva, I.; Atanassov, I.; Draganova, P.; Borisova, C.; Gärtner, G. Carotenoids in five aeroterrestrial strains from Vischeria/Eustigmatos group: Updating the pigment patterns of Eustigmatophyceae. Biotechnol. Biotechnol. Equip. 2019, 33, 250–267.
63. Huss, V.A.; Ciniglia, C.; Cennamo, P.; Cozzolino, S.; Pinto, G.; Pollio, A. Phylogenetic relationships and taxonomic position of Chlorella-like isolates from low pH environments (pH < 3.0). BMC Evol. Biol. 2002, 2, 13.
64. Darienko, T.; Pröschold, T. Genetic variability and taxonomic revision of the genus Auxenochlorella (Shihira et Krauss) Kalina et Puncocharova (Trebouxiophyceae, Chlorophyta). J. Phycol. 2015, 51, 394–400.
65. Gao, C.; Wang, Y.; Shen, Y.; Yan, D.; He, X.; Dai, J.; Wu, Q. Oil accumulation mechanisms of the oleaginous microalga Chlorella protothecoides revealed through its genome, transcriptomes, and proteomes. BMC Genomics 2014, 15, 582.
66. Shi, X.-M.; Liu, H.-J.; Zhang, X.-W.; Chen, F. Production of biomass and lutein by Chlorella protothecoides at various glucose concentrations in heterotrophic cultures. Process Biochem. 1999, 34, 341–347.
67. Shi, X.M.; Chen, F. Production and rapid extraction of lutein and the other lipid-soluble pigments from Chlorella protothecoides grown under heterotrophic and mixotrophic conditions. Nahrung 1999, 43, 109–113.
68. Shi, X.-M.; Chen, F.; Yuan, J.P.; Chen, H. Heterotrophic production of lutein by selected Chlorella strains. J. Appl. Phycol. 1997, 9, 445–450.
69. Heredia-Arroyo, T.; Wei, W.; Hu, B. Oil accumulation via heterotrophic/mixotrophic Chlorella protothecoides. Appl. Biochem. Biotechnol. 2010, 162, 1978–1995.
70. Gupta, A.K.; Seth, K.; Maheshwari, K.; Baroliya, P.K.; Meena, M.; Kumar, A.; Vinayak, V.; Harish. Biosynthesis and extraction of high-value carotenoid from algae. Front. Biosci. Landmark 2021, 26, 171–190.
71. Bhosale, P.; Bernstein, P.S. Microbial xanthophylls. Appl. Microbiol. Biotechnol. 2005, 68, 445–455.
72. Theriault, R.J. Heterotrophic growth and production of xanthophylls by Chlorella pyrenoidosa. Appl. Microbiol. 1965, 13, 402–416.
73. Cha, K.H.; Lee, H.J.; Koo, S.Y.; Song, D.-G.; Lee, D.-U.; Pan, C.-H. Optimization of pressurized liquid extraction of carotenoids and chlorophylls from Chlorella vulgaris. J. Agric. Food Chem. 2010, 58, 793–797.
74. Leya, T.; Rahn, A.; Lütz, C.; Remias, D. Response of arctic snow and permafrost algae to high light and nitrogen stress by changes in pigment composition and applied aspects for biotechnology. FEMS Microbiol. Ecol. 2009, 67, 432–443.
75. Remias, D.; Lütz-Meindl, U.; Lütz, C. Photosynthesis, pigments and ultrastructure of the alpine snow alga Chlamydomonas nivalis. Eur. J. Phycol. 2005, 40, 259–268.
76. Remias, D.; Lütz, C. Characterisation of esterified secondary carotenoids and of their isomers in green algae: A HPLC approach. Algol. Stud. 2005, 124, 85–94.

77. Sorigué, D.; Légeret, B.; Cuiné, S.; Morales, P.; Mirabella, B.; Guédeney, G.; Li-Beisson, Y.; Jetter, R.; Peltier, G.; Beisson, F. Microalgae synthesize hydrocarbons from long-chain fatty acids via a light-dependent pathway. Plant Physiol. 2016, 171, 2393–2405.

78. Giroud, C.; Gerber, A.; Eichenberger, W. Lipids of Chlamydomonas reinhardtii. Analysis of molecular species and intracellular site(s) of biosynthesis. Plant Cell Physiol. 1988, 29, 587–595.

79. Voshall, A.; Christie, N.T.M.; Rose, S.L.; Khasin, M.; Van Etten, J.L.; Markham, J.E.; Riekhof, W.R.; Nickerson, K.W. Sterol Biosynthesis in four green algae: A bioinformatic analysis of the ergosterol versus phytosterol decision point. J. Phycol. 2021, 57, 1199–2111.

80. Sasso, S.; Pohnert, G.; Lohr, M.; Mittag, M.; Hertweck, C. Microalgae in the post-genomic era: A blooming reservoir for new natural products. FEMS Microbiol. Rev. 2013, 36, 761–785.

81. John, U.; Beszteri, B.; Derelle, E.; Van de Peer, Y.; Read, B.; Moreau, H.; Cembella, A. Novel insights into evolution of protistan polyketide synthases through phylogenomic analysis. Protist 2008, 159, 21–30.

82. Remias, D.; Pichrtová, M.; Pangratz, M.; Lütz, C.; Holzinger, A. Ecophysiology, secondary pigments and ultrastructure of Chlainomonas sp. (Chlorophyta) from the European Alps compared with Chlamydomonas nivalis forming red snow. FEMS Micorbiol. Ecol. 2016, 92, fiw030.

83. Becker, E.W. Microalgae: Biotechnology and Microbiology; Cambridge University Press: Cambridge, UK, 1994.

84. Lee, R.E. Phycology, 4th ed.; Cambridge University Press: New York, NY, USA, 2008.

85. Yasukawa, K.; Akihisa, T.; Kanno, H.; Kaminaga, T.; Izumida, M.; Sakoh, T.; Tamura, T.; Takido, M. Inhibitory effects of sterols isolated from Chlorella vulgaris on 12-O-tetradecanoylphorbol-13-acetate-Induced inflammation and tumor promotion in mouse skin. Biol. Pharm. Bull. 1996, 19, 573–576.

86. Ambati, R.R.; Gogisetty, D.; Aswathanarayana, R.G.; Ravi, S.; Bikkina, P.N.; Bo, L.; Yuepeng, S. Industrial potential of carotenoid pigments from microalgae: Current trends and future prospects. Crit. Rev. Food Sci. Nutr. 2019, 59, 1880–1902.

87. Ambati, R.R.; Phang, S.-M.; Ravi, S.; Aswathanarayana, R.G. Astaxanthin: Sources, extraction, stability, biological activities and its commercial applications—A review. Mar. Drugs 2014, 12, 128–152.

88. Zhang, D.H.; Lee, Y.K. Two-step process for ketocarotenoid production by a green alga, Chlorococcum sp. strain MA-1. Appl. Microbiol. Biotechnol. 2001, 55, 537–540.

89. Zhang, D.H.; Lee, Y.K. Enhanced accumulation of secondary carotenoids in a mutant of the green alga, Chlorococcum sp. J. Appl. Phycol. 1997, 9, 459–463.

90. Zhang, D.H.; Ng, M.L.; Phang, S.M. Composition and accumulation of secondary carotenoids in Chlorococcum sp. J. Appl. Phycol. 1997, 9, 147–155.

91. Li, H.B.; Chen, F. Preparative Isolation and purification of astaxanthin from the green microalga Chlorococcum sp. by high-speed counter-current chromatography. In Algae and Their Biotechnological Potential; Chen, F., Jiang, Y., Eds.; Springer: Dordrecht, The Netherlands, 2001.

92. Masojídek, J.; Torzillo, G.; Kopecký, J.; Koblížek, M.; Nidiaci, L.; Komenda, J.; Lukavská, A.; Sacchi, A. Changes in chlorophyll fluorescence quenching and pigment composition in the green alga Chlorococcum sp. grown under nitrogen deficiency and salinity stress. J. Appl. Phycol. 2000, 12, 417–426.

93. Lang, I.; Hodač, L.; Friedl, T.; Feussner, I. Fatty acid profiles and their distribution patterns in microalgae: A comprehensive analysis of more than 2000 strains from the SAG culture collection. BMC Plant Biol. 2011, 11, 124.

94. Remias, D.; Wastian, H.; Lütz, C.; Leya, T. Insight into the biology and phylogeny of Chloromonas polyptera (Chlorophyta), an alga causing orange snow in Maritime Antarctica. Antarct. Sci. 2013, 25, 648–656.

95. Hoham, R.; Remias, D. Snow and glacial algae: A review. J. Phycol. 2020, 56, 264–282.

96. Koo, S.Y.; Cha, K.H.; Song, D.-G.; Chung, D.; Pan, C.-H. Optimization of pressurized liquid extraction of zeaxanthin from Chlorella ellipsoidea. J. Appl. Phycol. 2012, 24, 725–730.

97. Lutz, S.; Anesio, A.M.; Raiswell, R.; Edwards, A.; Newton, R.J.; Gill, F.; Benning, L.G. The biogeography of red snow microbiomes and their role in melting arctic glaciers. Nat. Commun. 2016, 7, 11968.

98. Procházková, L.; Remias, D.; Řezanka, T.; Nedbalová, L. Chloromonas nivalis subsp. tatrae, subsp. nov. (Chlamydomonadales, Chlorophyta): Re–examination of a snow alga from the High Tatra Mountains (Slovakia). Fottea 2018, 18, 1–18.

99. Spijkerman, E.; Wacker, A.; Weithoff, G.; Leya, T. Elemental and fatty acid composition of snow algae in Arctic habitats. Front. Microbiol. 2012, 3, 380.

100. Bar, E.; Rise, M.; Vishkautsan, M.; Arad, S. Pigment and structural changes in Chlorella zofingiensis upon light and nitrogen stress. J. Plant Physiol. 1995, 146, 527–534.

101. Orosa, M.; Torres, E.; Fidalgo, P.; Abalde, J. Production and analysis of secondary carotenoids in green algae. J. Appl. Phycol. 2000, 12, 553–556.

102. Pelah, D.; Sintov, A.; Cohen, E. The effect of salt stress on the production of canthaxanthin and astaxanthin by Chlorella zofingiensis grown under limited light intensity. World J. Microbiol. Biotechnol. 2004, 20, 483–486.

103. Ip, P.F.; Chen, F. Production of astaxanthin by the green microalga Chlorella zofingiensis in the dark. Process Biochem. 2005, 40, 733–738.

104. Ip, P.F.; Wong, K.-H.; Chen, F. Enhanced production of astaxanthin by the green microalga Chlorella zofingiensis in mixotrophic culture. Process Biochem. 2004, 39, 1761–1766.

105. Del Campo, J.A.; Rodriguez, H.; Moreno, J.; Vargas, M.A.; Rivas, J.; Guerrero, M.G. Accumulation of astaxanthin and lutein in Chlorella zofingiensis (Chlorophyta). Appl. Microbiol. Biotechnol. 2004, 64, 848–854.

106. Ye, Y.; Huang, J.-C. Defining the biosynthesis of ketocarotenoids in Chromochloris zofingiensis. Plant Divers. 2020, 42, 61–66.

107. Liu, J.; Sun, Z.; Gerken, H.; Liu, Z.; Jiang, Y.; Chen, F. Chlorella zofingiensis as an alternative microalgal producer of astaxanthin: Biology and industrial potential. Mar. Drugs 2014, 12, 3487–3515.

108. Mulders, K.J.M.; Weesepoel, Y.; Bodenes, P.; Lamers, P.P.; Vincken, J.-P.; Martens, D.E.; Gruppen, H.; Wijffels, R.H. Nitrogen-depleted Chlorella zofingiensis produces astaxanthin, ketolutein and their fatty acid esters: A carotenoid metabolism study. J. Appl. Phycol. 2015, 27, 125–140.

109. Orosa, M.; Valero, J.; Herrero, C. Comparison of the accumulation of astaxanthin in Haematococcus pluvialis and other green microalgae under N-starvation and high light conditions. Biotechnol. Lett. 2001, 23, 1079–1085.

110. Deenu, A.; Naruenartwongsakul, S.; Kim, S.M. Optimization and economic evaluation of ultrasound extraction of lutein from Chlorella vulgaris. Biotechnol. Bioprocess Eng. 2013, 18, 1151–1162.

111. Osipova, S.; Dudareva, L.; Bondarenko, N.; Nasarova, A.; Sokolova, N.; Obolkina, L.; Glyzina, O.; Timoshkin, O. Temporal variation in fatty acid composition of Ulothrix zonata (Chlorophyta) from ice and benthic communities of Lake Baikal. Phycologia 2009, 48, 130–135.

112. Abe, K.; Hattori, H.; Hirano, M. Accumulation and antioxidant activity of secondary carotenoids in the aerial microalga Coelastrella striolata var. multistriata. Food Chem. 2007, 100, 656–661.

113. Pollio, A.; Della Greca, M.; Monaco, P.; Pinto, G.; Previtera, L. Lipid composition of the acidophilic alga Dunaliella acidophila (Volvocales, Chlorophyta) I. Nonpolar lipids. Biochim. Biophys. Acta 1988, 963, 53–60.

114. De los Reyes, C.; Ávila-Román, J.; Ortega, M.J.; de la Jara, A.; García-Mauriño, S.; Motilva, V.; Zubía, E. Oxylipins from the microalgae Chlamydomonas debaryana and Nannochloropsis gaditana and their activity as TNF-α inhibitors. Phytochemistry 2014, 102, 152–161.

115. Fernández-Sevilla, J.M.; Acién-Fernández, F.; Molina-Grima, E. Biotechnological production of lutein and its applications. Appl. Microbiol. Biotechnol. 2010, 86, 27–40.

116. Bux, F. (Ed.) Biotechnological Applications of Microalgae Biodiesel and Value-Added Products; CRC Press: Boca Raton, FL, USA; Taylor & Francis Group: Abingdon, UK, 2013.

117. Del Campo, J.A.; Rodríguez, H.; Moreno, J.; Varga, M.Á.; Rivas, J.; Guerrero, M.G. Carotenoid content of chlorophycean microalgae: Factors determining lutein accumulation in Muriellopsis sp. (Chlorophyta). J. Biotechnol. 2000, 76, 51–59.

118. Del Campo, J.A.; Rodríguez, H.; Moreno, J.; Varga, M.Á.; Rivas, J.; Guerrero, M.G. Lutein production by Muriellopsis sp. in an outdoor tubular photobiorector. J. Biotechnol. 2001, 81, 289–295.

119. Del Campo, J.A.; García-González, M.; Guerrero, M.G. Outdoor cultivation of microalgae for carotenoid production: Current state and perspectives. Appl. Microbiol. Biotechnol. 2007, 74, 1163–1174.

120. Bigogno, C.; Khozin-Goldberg, I.; Boussiba, S.; Vonshak, A.; Cohen, Z. Lipid and fatty acid composition of the green oleaginous alga Parietochloris incisa, the richest plant source of arachidonic acid. Phytochemistry 2002, 60, 497–503.

121. Abu-Ghosh, S.; Pal-Nath, D.; Markovitch, D.; Solovchenko, A.; Didi-Cohen, S.; Portugal, I.; Khozin-Goldberg, I.; Cohen, Z.; Boussiba, S. A novel source of dihomo-γ-linolenic acid: Possibilities and limitations of DGLA production in the high-density cultures of the Δ5 desaturase-mutant microalga Lobosphaera incisa. Eur. J. Lipid Sci. Technol. 2015, 117, 760–766.

122. Lee, S.; Lim, S.R.; Jeong, D.G.; Kim, J.H. Characterization of an oleaginous unicellular green microalga, Lobosphaera incisa (Reisigl, 1964) Strain K-1, isolated from a tidal flat in the Yellow Sea, Republic of Korea. Front. Microbiol. 2018,

9, 2159.

123. Bigogno, C.; Khozin-Goldberg, I.; Cohen, Z. Accumulation of arachidonic acid-rich triacylglycerols in the microalga Parietochloris incisa (Trebuxiophyceae, Chlorophyta). Phytochemistry 2002, 60, 135–143.
124. Satoh, A.; Kato, M.; Yamato, K.; Ishibashi, M.; Sekiguchi, H.; Kurano, N.; Miyachi, S. Characterization of the lipid accumulation in a new microalgal species, Pseudochoricystis ellipsoidea (Trebouxiophyceae). J. Jpn. Inst. Energy 2010, 89, 909–913.
125. Procházková, L.; Leya, T.; Křížková, H.; Nedbalová, L. Sanguina nivaloides and Sanguina aurantia gen. et spp. nov. (Chlorophyta): The taxonomy, phylogeny, biogeography and ecology of two newly recognised algae causing red and orange snow. FEMS Microbiol. Ecol. 2019, 95, fiz064.
126. Chan, M.-C.; Ho, S.-H.; Lee, D.-J.; Chen, C.-Y.; Huang, C.-C.; Chang, J.-S. Characterization, extraction and purification of lutein produced by an indigenous microalga Scenedesmus obliquus CNW-N. Biochem. Eng. J. 2013, 78, 24–31.
127. Higuera-Ciapara, I.; Felix-Valenzuela, L.; Goycoolea, F.M. Astaxanthin: A review of its chemistry and applications. Crit. Rev. Food Sci. Nutr. 2006, 46, 185–196.
128. Remias, D.; Albert, A.; Lütz, C. Effects of realistically simulated, elevated UV irradiation on photosynthesis and pigment composition of the alpine snow alga Chlamydomonas nivalis and the arctic soil alga Tetracystis sp. (Chlorophyceae). Photosynthetica 2010, 48, 269–277.
129. Da Silva, M.E.T.; Martins, M.A.; de Oliveira Leite, M.; Milião, G.L.; dos Reis Coimbra, J.S. Microalga Scenedesmus obliquus: Extraction of bioactive compounds and antioxidant activity. Rev. Cienc. Agron. 2021, 52, e20196848.
130. Da Silva, M.E.T.; de Paula Correa, K.; Martins, M.A.; da Matta, S.L.P.; Martino, H.S.D.; dos Reis Coimbra, J.S. Food safety, hypolipidemic and hypoglycemic activities, and in vivo protein quality of microalga Scenedesmus obliquus in Wistar rats. J. Funct. Foods 2020, 65, 103711.
131. Rocha, D.N.; Martins, M.A.; Soares, J.; Vaz, M.G.M.V.; de Oliveira Leite, M.; Covell, L.; Mendes, L.B.B. Combination of trace elements and salt stress in different cultivation modes improves the lipid productivity of Scenedesmus spp. Bioresour. Technol. 2019, 289, 121644.
132. Wiltshire, K.H.; Boersma, M.; Möller, A.; Buhtz, H. Extraction of pigments and fatty acids from the green alga Scenedesmus obliquus (Chlorophyceae). Aquatic Ecol. 2000, 34, 119–126.
133. Qin, S.; Liu, G.-X.; Hu, Z.-Y. The accumulation and metabolism of astaxanthin in Scenedesmus obliquus (Chlorophyceae). Process Biochem. 2008, 43, 795–802.
134. Ho, S.H.; Chan, M.C.; Liu, C.C.; Chen, C.Y.; Lee, W.L.; Lee, D.J.; Chang, J.S. Enhancing lutein productivity of an indigenous microalga Scenedesmus obliquus FSP-3 using light-related strategies. Bioresour. Technol. 2014, 152, 275–282.
135. Thiyagarasaiyar, K.; Goh, B.H.; Jeon, Y.J.; Yow, Y.Y. Algae Metabolites in Cosmeceutical: An Overview of Current Applications and Challenges. Mar Drugs 2020, 18, 323.
136. Hofbauer, W.K. Toxic or otherwise harmful algae and the built environment. Toxins 2021, 13, 465.