

Review Article

# Recycle, repair, recover: the role of autophagy in modulating skeletal muscle repair and post-exercise recovery

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Skeletal muscle is a highly plastic tissue that can adapt relatively rapidly to a range of stimuli. In response to novel mechanical loading, e.g. unaccustomed resistance exercise, myofibers are disrupted and undergo a period of ultrastructural remodeling to regain full physiological function, normally within 7 days. The mechanisms that underpin this remodeling are believed to be a combination of cellular processes including ubiquitin-proteasome/calpain-mediated degradation, immune cell infiltration, and satellite cell proliferation/differentiation. A relatively understudied system that has the potential to be a significant contributing mechanism to repair and recovery is the autophagolysosomal system, an intracellular process that degrades damaged and redundant cellular components to provide constituent metabolites for the resynthesis of new organelles and cellular structures. This review summarizes our current understanding of the autophagolysosomal system in the context of skeletal muscle repair and recovery. In addition, we also provide hypothetical models of how this system may interact with other processes involved in skeletal muscle remodeling and provide avenues for future research to improve our understanding of autophagy in human skeletal muscle.

## Introduction

During recovery from resistance exercise (RE), mechanically perturbed myofibers undergo rapid ultrastructural remodeling to regain full physiological function normally within a week [1,2]. Successive bouts of RE accustoms skeletal muscle to loading [3,4] and significant myofibrillar protein accrual (i.e. fiber cross-sectional area growth) can be observed after 10–12 weeks of resistance training (RT) [5–7]. This hypertrophic response is fundamentally driven by the combined effect of incremental mechanical loading and consistent dietary amino acid availability, which increases muscle protein synthesis (MPS) beyond muscle protein breakdown (MPB) for positive net protein balance over time [8,9]. The initial protein synthetic increase during early-stage RT is, however, likely indicative of a global stress response to novel exercise-induced muscle damage (EIMD) rather than hypertrophy adaptation *per se* [6,10–12]. While the necessity of this initial damage response for muscle hypertrophy has been debated [5,13,14], unaccustomed eccentric exercise evokes a degree of ultrastructural deformation and functional impairment [1], which likely needs to be attenuated for adaptive remodeling to ensue [6]. Therefore, identifying the mechanisms underpinning skeletal muscle recovery may uncover potential methods to enhance athletic performance or expedite training-induced adaptations.

While most post-exercise recovery strategies favor the stimulation of skeletal muscle anabolism, recent evidence suggests that changes in myofibrillar MPS do not directly explain improved exercise recovery when dietary protein is sufficient [15]. Further, an inability to synthesize myofibrillar proteins does not seem to be the underlying cause of delayed skeletal muscle recovery in aging rodents [16]. These intriguing data could imply that, in the context of muscle damage, MPB could be a modulating factor. However, despite the increasing need to consider proteostatic mechanisms holistically [16–18], our current understanding of human skeletal muscle catabolism is poor relative to anabolism [19]. Intracellular degradation is predominantly regulated by the calpain, ubiquitin-proteasome (UPS), and autophagolysosomal systems, each of which has their own distinct underpinning signaling pathways. Throughout this review, we will focus on the role of the autophagolysosomal system in skeletal muscle, with particular emphasis on its (potential) role during recovery from EIMD. For an in-depth overview of the

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role of the calpain and UPS systems in skeletal muscle, readers are directed to the review provided by Goll et al. [20].

## Overview of autophagy

Autophagy, meaning ‘self-eating’, is a conserved intracellular process originally conceptualized in 1963 by the discoverer of the lysosome, De Duve [21], and mechanistically expanded upon in yeast by Ohsumi [22,23] during the 1990s. Today, it is evident that autophagy is a pivotal quality control mechanism through which mammalian cells maintain tissue homeostasis under basal conditions [24,25] and in response to physiological stress [25,26]. Unlike the protein-specific UPS, autophagy also facilitates the degradation of lipids, carbohydrates, and nucleotides via acid hydrolysis. An energy-depleted and/or metabolically perturbed intracellular environment initiates the autophagic process, allowing redundant structures to be recycled into their constituent metabolites for anabolic (i.e. growth of cellular components) or catabolic (i.e. adenosine triphosphate resynthesis) repurposing. Abnormal clearance of such components can be detrimental to skeletal muscle health; thus, it is unsurprising aberrant autophagy has been linked to a variety of muscular diseases, the primary category being lysosomal storage disorders [27].

There are three known types of autophagy distinguishable by the method of cargo delivery to the lysosome: micro-autophagy, chaperone-mediated autophagy, and macro-autophagy. Although likely interconnected [28], a holistic discussion of these pathways is beyond the scope of this review; therefore, readers are referred elsewhere for overviews of chaperone-mediated [29] and micro-autophagy [30]. Macro-autophagy, henceforth referred to as autophagy, involves the intricate and highly coordinated interaction of autophagolysosomal machinery. Here, damaged or redundant organelles/cytosolic constituents are engulfed by nascent double-membrane autophagosome vesicles, which, in turn, fuse with lysosomes where the isolated cargo is catabolized. Originally thought to be an entirely non-selective process, more than 30 selective autophagy receptors have now been discovered [31] displaying how this pathway acts not only as a global stress response but can also target specific intracellular components.

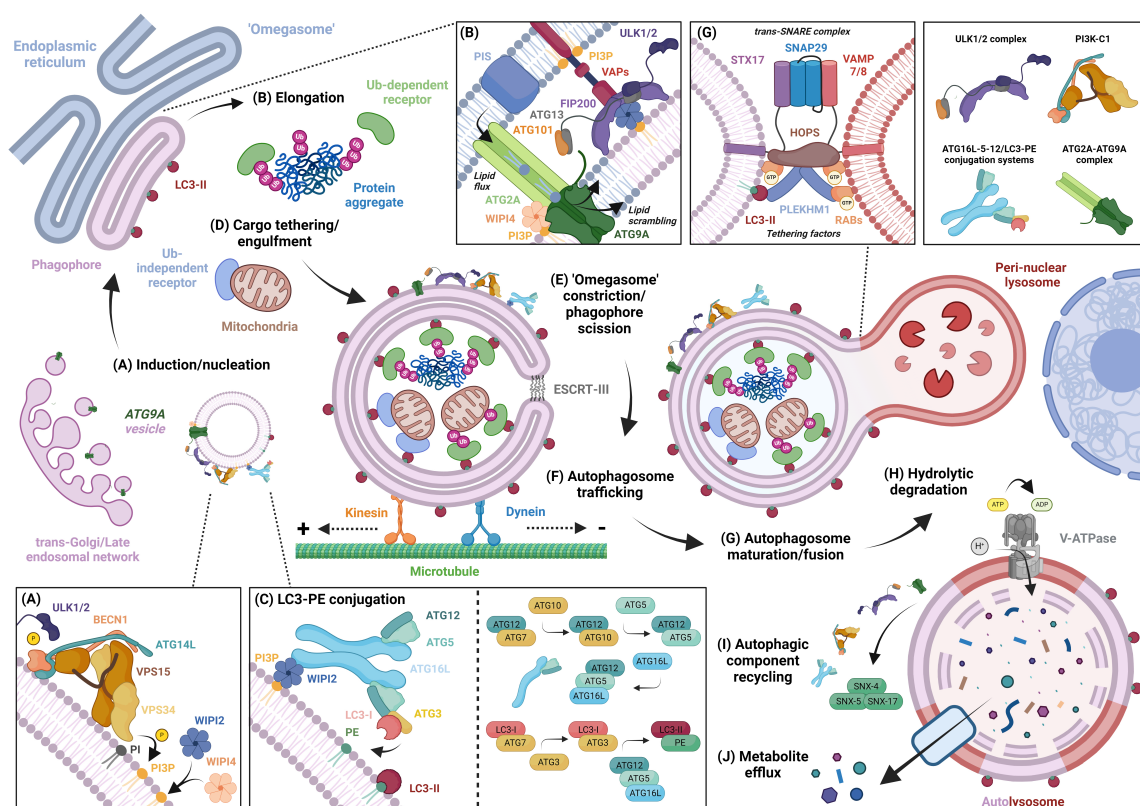
## Molecular mechanism of autophagosome biogenesis and degradation

The autophagic process is complex but can be broken down into four distinct stages: (1) induction and nucleation of the pre-autophagosome phagophore, (2) expansion of the phagophore membrane, (3) autophagosome maturation/lysosome fusion, and (4) hydrolytic degradation and efflux of metabolites.

Autophagic induction and phagophore nucleation are primarily coordinated by the Unc51-like kinase 1/2 (ULK1/2) autophagy initiation complex, comprising the ULK1/2 kinase, autophagy-related gene (ATG) 13, ATG101, and scaffold protein focal adhesion kinase family interacting protein of 200 kD (FIP200), and the autophagy specific class III phosphatidylinositol 3-kinase complex 1 (PI3K-C1), containing the phosphatidylinositol 3-phosphate (PI3P) kinase vacuolar protein sorting (VPS) 34, VPS15, ATG14L, and Beclin-1. Upon activation, ULK1/2 complexes are recruited to the phagophore initiation site where they exert kinase activity toward PI3K-C1 for PI3P production (Figure 1A) [32,33]. Localized accumulation of PI3P at the initiation site recruits WD repeat domain (WIPI) PI3P-binding proteins, triggering the assembly of downstream autophagy machinery for membrane elongation [34].

Mammalian autophagosomes are predominantly formed in association with an endoplasmic reticulum (ER) subdomain with an  $\Omega$ -like shape, fittingly termed the ‘omegasome’ [35]. Here, phagophore-ER contact sites are established between integrated ER VAMP-associated proteins (VAPs) and FIP200/ULK1 in a PI3P-dependent manner [36], while ATG2A and ATG9A transfer lipids from the ER to the elongating phagophore membrane (Figure 1b) [37,38]. ATG9A is trafficked to the phagophore assembly site within Golgi/endosome-derived vesicles [39,40], which form the initial phagophore seed [41] and begin to accumulate autophagic machinery [33]. The ubiquitin-like ATG16L-ATG5-ATG12 conjugation system associates with the growing phagophore to convert cytoplasmic microtubule-associated protein 1A/1B-light chain 3 (LC3-I) into membrane-bound phosphatidylethanolamine (PE) conjugated LC3-II [34,42–45] in a series of reactions involving ATG10, ATG7, and ATG3 (Figure 1C) [46–49].

LC3 isoforms and their gamma-aminobutyric acid receptor-associated protein (GABARAP) subfamily are structurally like other ubiquitin-like proteins but contain two extra  $\alpha$ -helices that act as docking regions for autophagy-related proteins [50]. Several ATGs within the ULK1/2 complex, PI3K-C1, and ATG2A



**Figure 1: General mechanism of autophagosome biogenesis and degradation.**

(A) Activated ULK1/2 complexes localize at the phagophore induction site and phosphorylates Beclin-1, allowing VPS34 to convert PI to PI3P for recruitment of WIPI proteins. (B) Recently nucleated phagophores are tethered to the 'Omegasome' by interactions between integral ER VAPs, ULK1/FIP200, and WIPI2. ATG2A associates with ATG9A via WIPI4 and transfers synthesized ER lipids to the elongating phagophore, which are equilibrated by the ATG2A lipid scramblase. (C) LC3/GABARAP proteins are embedded into the autophagic membrane through a series of ubiquitin-like reactions mediated by ATG7 and ATG3 as well as the ATG12-5-16L complex. (D) Autophagic cargo is tethered to LC3-II on the nascent phagophore membrane through connecting autophagy receptors. (E) Fully elongated phagophores are sealed by ESCRT machinery and dissociate from their ER donor through Omegasome constriction. (F) Newly formed autophagosomes are transported along microtubules by kinesin or dynein motor proteins, where they eventually encounter lysosomes. (G) GTP-loaded Rab proteins and LC3-II promote the tethering of the lysosomal and outer autophagosome membrane through PLEKHM1 and its HOPS complex effector allowing SNARE-mediated fusion of the membrane structures. (H) The V-ATPase acidifies the autolysosome lumen allowing hydrolysis of the inner autophagosome membrane, connecting autophagy adaptors, and sequestered cargo. (I) Autophagic components are extracted from the autolysosome membrane by the SNX4-5-17 recycler complex and degraded materials are released into the cytosol (J). Created in BioRender. Acheson, J. (2024) BioRender.com/a02s604.

lipid-transferase harbor LC3-interacting regions; thus, it is thought that lipidated LC3 and GABARAP accelerate phagophore expansion by providing additional scaffolding sites for autophagic machinery [51–53]. Furthermore, during selective autophagy, LC3/GABARAP mediates tethering of predetermined cargo to the inner phagophore membrane (Figure 1D). Ubiquitin-sensitive autophagy receptors, such as sequestosome-1 (p62), connect ubiquitinated cargo to PE-conjugated LC3/GABARAP through LC3 and ubiquitin-binding domains [54], whereas ubiquitin-independent receptors such as BCL2/Adenovirus E1B 19 kDa protein-interacting protein 3 (BNIP3) and BNIP3-like (BNIP3L/NIX), directly bind cargo to LC3/GABARAP [55,56].

Autophagosomes are formed once fully elongated phagophores undergo endosomal sorting complexes required for transport (ESCRT) mediated membrane scission [57–59] and omegasome constriction, allowing the autophagosome to dissociate from its membrane donor (Figure 1E) [60,61]. Newly formed autophagosomes are then bound to motor scaffold proteins and transported along the microtubule network via dynein [62–64] and kinesin [65] toward juxtanuclear lysosomes (Figure 1F) [66]. Here, concerted actions between Rab guanosine triphosphatases (GTPases), LC3 proteins, homotypic fusion and protein sorting (HOPS)-tethering factors, and soluble N-ethylmaleimide-sensitive factor attachment protein receptors (SNARE) promote tethering and fusion of the outer autophagosome and lysosomal membranes (Figure 1G), subsequently creating an autolysosome [67–70].

Autophagic catabolism within autolysosomes involves the hydrolysis of the inner autophagosome membrane, the cargo it surrounds, and the connecting autophagy adaptors. Such degradation can be

carried out by over 60 lysosomal hydrolases [71], which require acidification of the autolysosome lumen by vacuolar H<sup>+</sup>-adenosine triphosphatases (v-ATPase) (Figure 1H). Importantly, however, recent evidence has suggested that proteins of the outer autophagosome membrane are recycled for future use (Figure 1I) [72,73]. Similarly, for efficient autophagy [74], lysosomal bodies must be extracted from the autolysosome through a separate process termed autophagic lysosome reformation (reviewed in-depth elsewhere [75]). Following catabolism within the autolysosome, constituent elements of degraded material are released into the cytosol through various transporters/channels for subsequent recycling into new cellular components (Figure 1J) [76].

The autophagolysosomal system is a highly regulated pathway and there are a myriad of stages that have the potential to be affected by stimuli such as mechanical disruption/damage of tissue. This is important as common markers of autophagosomes (e.g. LC3-II and p62) fluctuate depending on their rate of synthesis and autophagic degradation [77], meaning ‘static’ assessments of these proteins could misrepresent true rates of autophagy. Therefore, control comparators where autophagosome degradation has been inhibited (e.g. with colchicine) are required to confirm whether autophagy ‘flux’ (i.e. autophagosome synthesis and degradation) has increased or decreased in response to experimental conditions.

## Upstream regulation of autophagy

### Nutrient-sensitive autophagy signaling

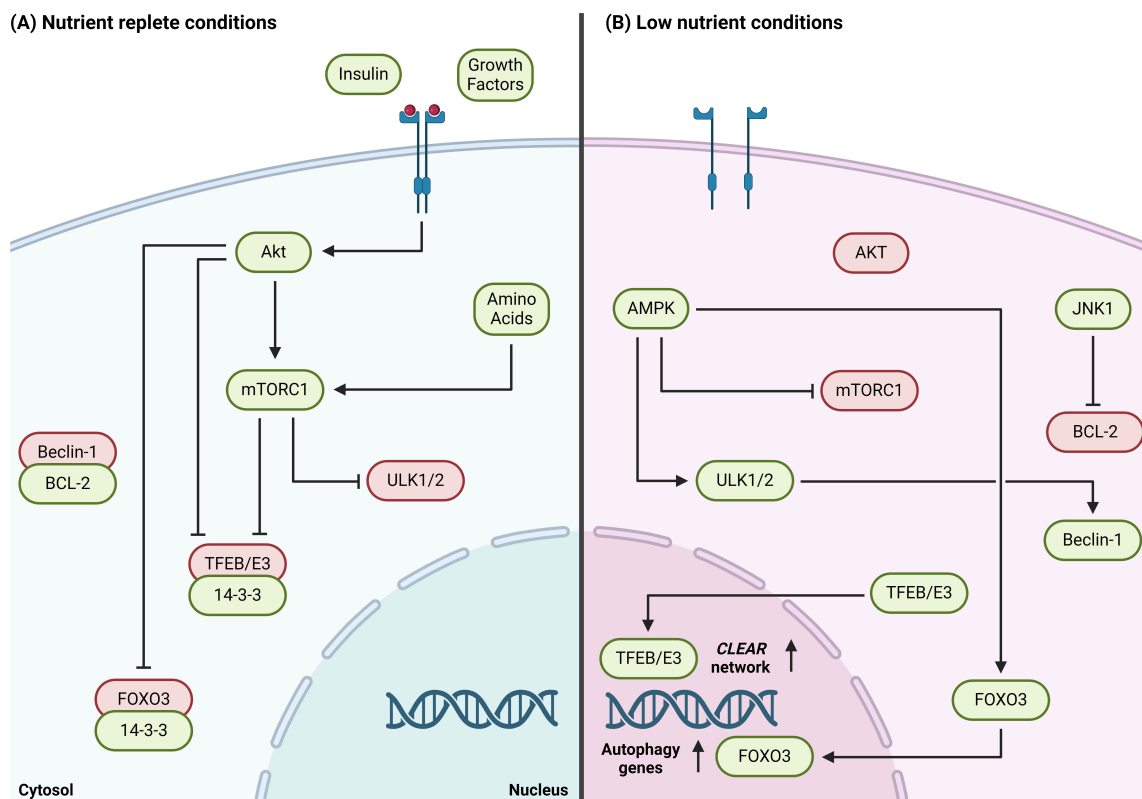
Our understanding of the upstream signals that regulate autophagy primarily stems from *in vitro* or animal studies utilizing nutrient withdrawal or pharmaceutical autophagy inhibitors. During periods where nutrients are replete, the active mammalian target of rapamycin complex 1 (mTORC1) inhibits the ULK1/2 complex by phosphorylating ULK1<sup>ser757</sup> and ATG13<sup>ser258</sup> [78–81]. Upstream of mTORC1, the growth factor/insulin-sensitive protein kinase B/AKT (AKT) phosphorylates transcription factor forkhead box O3 (FOXO3) at several inhibitory sites, promoting its interaction with cytosolic 14-3-3 to suppress autophagy gene transcription [82–85]. In a similar mechanism, both AKT and mTORC1 prevent coordinated lysosomal enhancement and regulation (CLEAR) gene network expression by phosphorylating members of the microphthalmia/transcription factor E (MiT/TFE) family, such as transcription factor EB (TFEB) and transcription factor binding to IGHM enhancer 3 (TFE3) [86–88].

In contrast, during nutrient deficiency, a reduction in mTORC1 signaling alleviates the induction complex’s negative inhibition on ULK1/2 and TFEB, allowing autophosphorylation of ATG13 and FIP2000 by ULK1/2 [79,80] and TFEB nuclear translocation for CLEAR gene network expression [89]. Meanwhile, an increase in cellular adenosine monophosphate (AMP):triphosphate (ATP) activates the energy sensor 5’ AMP-activated protein kinase (AMPK), which promotes autophagy induction by phosphorylating ULK1/2 at multiple serine residues [90–92] and elevates autophagy-related gene expression via phosphorylation of FOXO3 [92,93]. AMPK may also indirectly promote autophagy, given its ability to inhibit mTORC1 through phosphorylation of tuberous sclerosis protein and raptor [78,90–92]. Finally, phosphorylation of B-cell leukemia/lymphoma 2 protein (BCL-2) at several residues by starvation-activated c-Jun N-terminal protein kinase 1 (JNK1) attenuates BCL-2’s negative inhibition of Beclin-1 [94,95]. These opposing mechanisms allow cellular autophagy to be regulated temporally in response to changes in nutrient status (Figure 2).

### Redox/calcium-sensitive autophagy signaling

While there are various potential upstream autophagic regulators, disturbance of intracellular redox and calcium homeostasis has been strongly implicated in skeletal muscle autophagy (Figure 3) [96–98]. Mitochondrial-derived reactive oxygen species (ROS) are thought to be key regulators of contraction-induced autophagy induction [99–101]. Mechanistically, ROS activate AMPK by reducing cellular ATP [102], upregulate autophagy by attenuating AKT signaling [103,104], and directly oxidize MiT/TFE transcription factors to promote autophagolysosomal gene expression [105]. Furthermore, ROS stimulate lysosomal calcium release by oxidizing the lysosome calcium channel mucolipin-1/TRPML1 (MCOLN1) [106,107] which, in turn, promote TFEB nuclear translocation to elevate lysosomal/autophagy-related gene expression [108]. MCOLN1 also activates the AMPK-effector calcium/calmodulin-dependent protein kinase kinase  $\beta$  (CaMKK $\beta$ ), resulting in phosphorylation of ULK1 and Beclin-1 [109].





**Figure 2: Upstream regulators of nutrient-sensitive autophagy.**

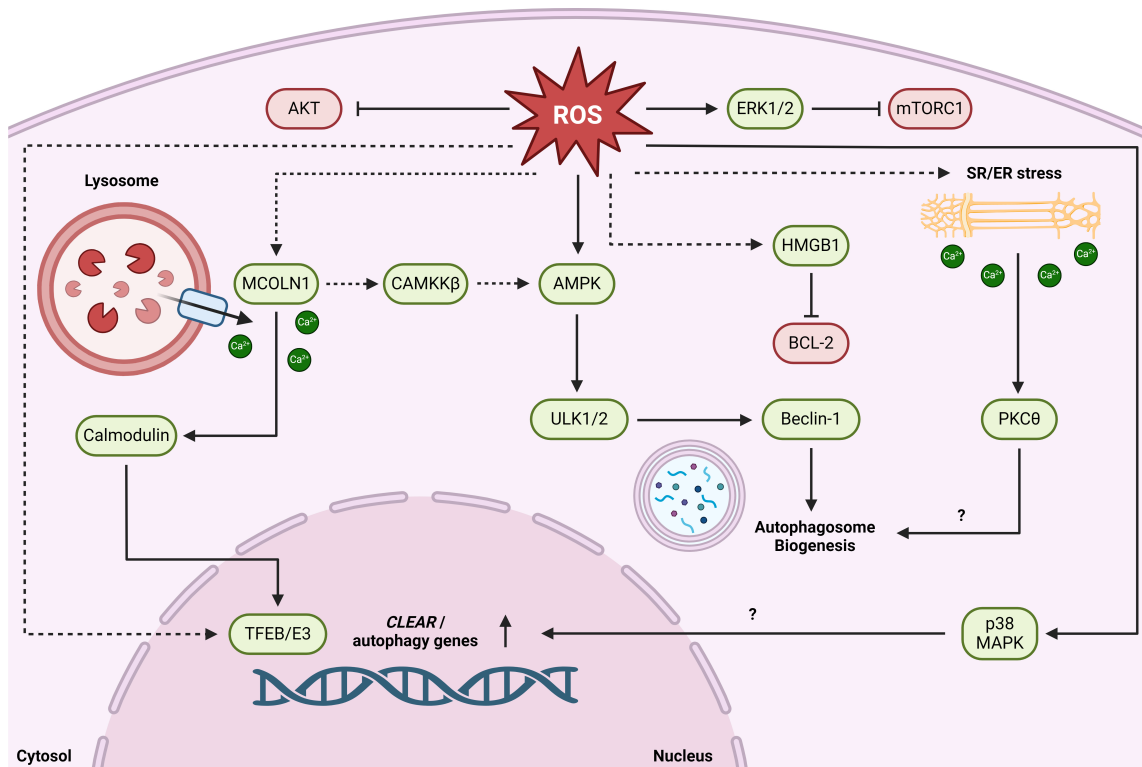
(A) During periods where growth factors and amino acids are replete, the AKT/mTORC1 pathway negatively regulates autophagy induction and transcriptional programs. mTORC1 directly phosphorylates and inhibits ULK1/2, ULK1/2, and ATG13. TFEB/E3 are phosphorylated by both AKT and mTORC1 and sequestered in the cytosol by 14-3-3 proteins, while AKT similarly phosphorylates and prevents FOXO3-mediated autophagy gene expression. Autophagosome biogenesis is suppressed through BCL-2-mediated inhibition of Beclin-1. (B) In low nutrient conditions, a reduction in AKT/mTORC1 signaling alleviates the negative inhibition of the ULK1/2 complex. The energy sensor AMPK simultaneously phosphorylates and activates ULK1/2, allowing it to phosphorylate Beclin-1, which has been released from BCL-2 by JNK1. FOXO3 and TFEB/E3 translocate to the nucleus and upregulate autophagy and CLEAR network gene expression due to the reduction in AKT/mTORC1 activity and direct phosphorylation of FOXO3 by AMPK. Created in BioRender. Acheson, J. (2024) BioRender.com/r89e937.

Alike starvation-induced autophagy, exercise promotes the phosphorylation and dissociation of BCL-2 from Beclin-1, albeit via a differing mechanism [110]. In addition, exercise-induced phosphorylation of p38 mitogen-activated protein kinases (MAPK) has been implicated in upregulating autophagy gene expression [111], which could reflect upstream regulation by oxidative stress [112] or inflammatory receptor activation [113]. Cytosolic calcium mobilization in response to sarcoplasmic reticulum (SR) stress [114,115] may also promote autophagosome formation via protein kinase C theta (PKCθ) [116], while ROS produced during lactate clearance stimulates phosphorylation of extracellular signal-regulated kinases 1/2 (ERK1/2), thereby inhibiting mTORC1 and promoting autophagy flux [117]. Collectively, these mechanistic studies highlight potential redox and calcium-sensitive signaling cascades that may be upregulated during recovery from strenuous exercise, albeit the degree to which such pathways influence autophagy flux in human skeletal muscle is yet to be determined.

## Exercise-induced muscle damage

### Defining exercise-induced muscle damage

It is well known that mechanical and/or metabolic stress during strenuous physical exercise causes temporary muscle damage and functional impairment [2,118–121]. Human experimental models of EIMD clearly show that repeated isolated eccentric muscle contractions significantly disrupt myofiber integrity and muscle force generating capacity [122–129], the most appropriate proxy of ultrastructural damage [130,131]. In severe cases of EIMD, evidence of myofiber necrosis, such as intramyofiber immune cell



**Figure 3: Redox and calcium-related autophagy signaling.**

Perturbations in cellular redox and intracellular calcium homeostasis can result in autophagy induction and related gene expression. ROS increase ULK1/2 complex activity through allosteric activation of AMPK and attenuation of AKT/mTORC1 activity. Lysosomal calcium efflux through MCOLN1, which can be induced by ROS, results in autophagy enhancement through CaMKKβ/AMPK activation and by activating TFEB via calmodulin. ROS may also promote autophagy gene expression through direct oxidation of TFEB/E3 and increasing p38 MAPK signaling. Oxidative stress results in sarcoplasmic reticulum efflux, associated with autophagosome biogenesis through activation of PKCθ. ROS promote HMGB1 relocation to the cytosol where it promotes autophagy induction by competitively binding BCL-2. Dashed lines represent autophagic pathways that remain to be documented in skeletal muscle. Created in BioRender. Acheson, J. (2024) BioRender.com/f77k718.

infiltration [127,129,132,133], particularly in dystrophin-negative myofibers [133], is also observed. Such effects are often preceded by a substantial (~50%) loss of muscle force generating capacity that requires > 7 days to recover [127,132,133]. In contrast, more traditional resistance and endurance-type exercise results in milder muscle strength losses and shorter recovery periods [134,135], although ultrastructural damage is noted following high-intensity RE [6,136–139] and downhill running [140,141], reflecting the eccentric demand of these modalities.

## Mechanisms of exercise-induced muscle damage

To understand how autophagy may contribute to skeletal muscle recovery, it is important to consider the etiology of EIMD. This can be divided into two distinct phases: the initial primary damage phase, relating to the immediate exercise-induced disruption of intracellular proteins and organelles, and the secondary damage phase, which occurs as an autogenic response to the initial damaging event.

### Primary damage

Eccentric loading of contractile proteins is likely the primary source of ultrastructural damage during weight-bearing exercise [142]. Compared to concentric loading, eccentric contractions recruit a lower number of motor units, which results in a greater amount of tensile stress per unit of muscle fiber area [143,144]. As a result, elongating sarcomeres progressively weaken and eventually stretch beyond myofilament overlap, subsequently placing a greater strain on surrounding structures (see Morgan's [145] 'popping sarcomere hypothesis'). In support of this theory, disruption of sarcomeres (e.g. Z-line/disk 'smearing' or myofilament disorganization), the t-tubule system, SR, and intermyofibrillar mitochondria

can all be observed in skeletal muscle tissue immediately after eccentric exercise [146–148]. Furthermore, much of the acute post-exercise reduction in muscle function has been attributed to a rise in intracellular calcium concentrations and subsequent excitation-contraction uncoupling [148,149]. While the precise mechanism of cytosolic calcium influx is debated [150], evidence from both human and animal investigations suggests that stretch-mediated and/or oxidative disruption of t-tubules, sarcolemma, and the SR may be involved [122,148,151–153].

## Secondary damage

During the secondary phase, a rise in cytosolic calcium levels signals the activation of calcium-sensitive calpains [154–156]. It is thought that these non-lysosomal proteases disassemble damaged myofibrillar and cytoskeletal structures, subsequently allowing the UPS to degrade unbound protein fragments into smaller peptide chains [20]. Indeed, human eccentric exercise-induced myofibrillar disruption and loss of muscle function directly correlate with calpain activity [155], while proteasome activity increases during the post-exercise period [157]. However, given that the presence of myofibrillar disruption is often delayed [155,158,159], changes in tissue ultrastructure during the days following exercise likely reflect a remodeling response to strenuous loading rather than further ‘damage’. Elevated cytosolic calcium may also increase mitochondrial calcium uptake and ROS formation [160], which are known to cause lipid, protein, and DNA oxidation in exercised-human muscle [161]. However, recent animal evidence has shown that transient ROS formed by localized mitochondrial calcium uptake are important for sarcolemma repair and maintaining myofiber viability following eccentric-damage, whereas sustained increases in cellular ROS hinder homeostatic regain [162]. Other rodent-based studies suggest that redox imbalances created by eccentric exercise may promote mitochondrial calcium overload and permeability [160,163,164], which increases the risk of pro-apoptotic factors entering the cytosol [165].

Inflammation is another key factor associated with secondary EIMD. Various cytokines are elevated in skeletal muscle and the circulation following eccentric exercise (for an extensive list, see Paulsen et al. [1]), some of which have been shown to coordinate the immune and myogenic response in cultured human skeletal myotubes [166–169]. Neutrophils initially accumulate in damaged human skeletal muscle tissue with pro-inflammatory M1-like phagocytic macrophages predominating soon after [133,167]. These phagocytic cells contribute to muscle healing by clearing cellular debris and further regulating the immune response, although cell culture and rodent models of muscle injury indicate that neutrophils also exhibit a role in generating cytolytic intermediates during phagocytosis [170–174]. Mouse M1 macrophages differentiate into a pro-regenerative, anti-inflammatory M2 phenotype upon engulfing muscle debris *in vitro* [175], and this timely expression seems to be important for regulating satellite cell dynamics and healthy myofiber regeneration in both humans and mice [174–178]. Importantly, however, a distinct change in macrophage phenotype may not occur in human skeletal muscle during recovery from traditional, less damaging forms of RE, possibly reflecting a lack of need to degrade necrotic tissue [179]. Nevertheless, macrophage and satellite cell accretion remain tightly coupled in response to exercise training [180–182]; thus, interactions between immune and myogenic cells are likely important for muscle recovery regardless of myofiber necrosis.

## Autophagy is essential for rodent skeletal muscle health and regeneration

Studies of skeletal muscle-specific ATG-knockout (KO) rodents have uncovered the pivotal role autophagy plays in maintaining myofiber homeostasis. Maserio et al. [24] delineated the importance of basal autophagy by generating life-long and tamoxifen-inducible ATG7-KO mouse lines. These animals were unable to lipidate LC3 and had substantial LC3 and p62 build-up, indicating a blockage of autophagosome removal. Both genotypes displayed loss of force production, indices of muscle damage, and skeletal muscle atrophy, which coincided with increased proteolytic gene expression and diminished activity of protein translational machinery, suggestive of a catabolic phenotype. ATG5-KO also induces glycolytic myofiber atrophy, which is associated with autophagy protein build-up and impaired lysosome morphology within the intermyofibrillar space [183]. Intriguingly, slow-twitch muscle and measures of muscle fatiguability were observed to be unaffected by ATG5-KO, highlighting a need to consider muscle-fiber type when investigating the autophagic response.

Pare et al. [184] showed that mouse chronic ATG7-KO hindered muscle force-production and measures of contractility in both the fast-twitch dominant extensor digitorum longus (EDL) and slow-twitch soleus muscle, but these effects were more pronounced and had an earlier onset in EDL. Analyses of autophagy-sufficient muscle showed that the EDL had higher basal autophagic flux and both basal and starvation-induced LC3-II accumulation are negatively associated with skeletal muscle citrate synthase activity [185], indicating that muscle with low oxidative capacity has greater autophagic turnover. Notably, glycolytic muscle is more susceptible to MPS attenuation and subsequent muscle loss during catabolic conditions [186,187], including aging where the decline in fast-twitch muscle size and function has been associated with dysfunctional autophagy [188]. However, preferential glycolytic muscle fiber wasting in sarcopenic muscle appears to be mTORC1 independent whereby inflammatory cytokines suppress autophagy via FYN/signal transducer and activator of transcription 3 signaling [189,190]. Nevertheless, the observation that fast-twitch muscle is especially reliant upon autophagy to maintain its health highlights important considerations in circumstances where glycolytic fibers are preferentially recruited, such as during eccentric loading [191].

In addition to basal autophagy, animal investigations indicate that the autophagy system plays a crucial role in recovering severely injured skeletal muscle [26,192,193]. Nichenko et al. [192] showed that suppressing autophagy significantly impeded the recovery of mitochondrial enzyme activity and muscle strength in mice exposed to localized cardio-toxin (Ctx) injury. This does not seem to be due to global autophagy repression as similar results have been reported in muscle-specific ULK1-KO mice [26]. Interestingly, ULK1-KO does not impede basal skeletal muscle health in young muscle [26], but does impair mitochondrial homeostasis and skeletal muscle contractility with advancing age [194]. Therefore, it seems ULK1-mediated autophagy facilitates skeletal muscle health and recovery during aging and acute skeletal muscle trauma in mice.

ATG16L-KO, which impedes, but does not entirely suppress autophagosome formation, also significantly delays muscle recovery from Ctx injury [195], with these mice exhibiting elevated sarcolemmal damage, smaller regenerating fibers and lower amounts of both proliferating and differentiating satellite cells compared to their autophagy-sufficient littermates. The effect on satellite cells seems to be of particular importance given that autophagy is upregulated during myoblast proliferation and differentiation *in vitro* [196–198]. Furthermore, loss of basal autophagy through satellite cell-specific ATG7-KO reduces the satellite cell pool in young mice, indicating that autophagy can prevent myogenic cell senescence/death [199]. Accordingly, it is believed that autophagy preserves satellite cell proteostasis and provides the necessary energy for mitosis [198,200]. Intriguingly, *myogenin-Cre* ablation of ULK1, which attenuates autophagy in postmitotic myofibers, represses the myogenic program during recovery from freeze injury, suggesting that autophagy within adult myofibers can regulate satellite cell dynamics [193]. Overall, these data show that autophagy facilitates rodent skeletal muscle repair/regeneration through both intrinsic and extrinsic (satellite cell) mechanisms and that these pathways are interrelated.

It is important to note that an increase in autophagy protein content may not translate to a relative increase in autophagic flux. Investigations conducted by Jarrod Call's laboratory have shown that while autophagy protein content is elevated in mouse skeletal muscle recovering from traumatic freeze injury, 2-photon microscopy analyses show a reduced clearance of autophagosome bound LC3, indicative of attenuated autophagosome clearance [193,201]. The authors postulated that this could be due to an autophagy 'bottleneck' whereby accumulating damaged components are not effectively degraded despite increases in autophagosome production [202]. One possible explanation for this may be that lysosomal biogenesis fails to proportionally increase with damage-induced autophagosome formation. For example, mouse cardiac muscle recovering from ischemia reperfusion (I/R) undergoes a robust increase in autophagosome formation yet is accompanied by a decline in the lysosomal marker lysosome-associated membrane protein 2 (LAMP2) [203], while restored expression of LAMP2 significantly improved the clearance of I/R induced autophagosomes [203]. Similar effects have been shown during TFEB overexpression in cultured I/R injured mouse cardiomyocytes [204], suggesting that a general increase in lysosomal number can overcome the 'bottleneck'. Overall, these data highlight the lysosomal system as a target to improve autophagic flux and subsequently skeletal muscle recovery.



## Autophagy is upregulated in rodent and cellular models of exercise-induced muscle damage

Salminen and Vihko's seminal work provided that the first indication that autophagy may be upregulated following EIMD [205], reporting that 9 hours of running causes significant myofiber necrosis and inflammatory cell infiltration in mouse quadriceps muscle. Surviving myofibers exhibited mitochondria-containing autophagic-like vacuoles at days 2 and 7 post-injury, indicative of increased autophagosome production. Using a similar model, Salminen and Kihlström [206] observed markers of lysosomal activity increased stepwise with greater dosages of exercise and subsequent tissue injury, suggesting that a more potent autophagic response occurs with higher magnitudes of EIMD. In more recent years, a growing body of research has identified a notable relationship between markers of autophagy and rodent mitochondrial dysfunction following EIMD [207–209]. Shang et al. [209] reported that 90 minutes of downhill running increased the LC3-II/LC3-I ratio and co-localization of PTEN-induced putative protein kinase 1 (PINK)/Parkin with dysfunctional mitochondria, for up to 48 hours post-exercise. The accumulation of PINK1 and its E3-ligase effector Parkin on depolarized mitochondrial membranes are key stages of ubiquitin-dependent mitophagy and are often preceded by mitochondrial fission events (see Erlich and Hood [210]). Mechanistically, it has recently been reported that the high mobility group box-1 protein (HMGB1), a structural component of chromatin [211], promotes autophagy induction during recovery from downhill running by translocating to the cytosol and relieving BCL-2's inhibition of Beclin-1 (Figure 3) [207]. A similar mechanism is noted in murine fibroblasts where mitochondrial ROS promote HMGB1-translocation and Beclin-1 mediated autophagy, whereas HMGB1 ablation impedes autophagy, resulting in apoptosis [212]. As such, HMGB1-translocation seems to be a key autophagic signal during conditions that stimulate mitochondrial stress (e.g. EIMD).

Maintaining a healthy mitochondrial pool is crucial for myofiber viability, especially during periods where intracellular calcium homeostasis is perturbed [213]. Investigations of mice with skeletal muscle dystrophy, where calcium [214] and autophagic [215] dysfunction are exhibited, illustrate that an inability to sequester damaged mitochondria exacerbates muscle degeneration and apoptosis during recovery from acute endurance exercise (EE) [216,217]. Mitochondrial turnover is likely important for general skeletal muscle recovery, given the significant energy cost of MPS [218,219]. Indeed, alike autophagy suppression [195,197], inhibition of mitochondrial biogenesis attenuates muscle regeneration following traumatic injury [220]. While the relationship between mitophagy and myogenesis is yet to be explored in the context of EIMD, mitochondrial oxidative stress peaks 6 hours after strenuous EE and coincides with elevated markers of mitophagy, suggesting that mitochondrial turnover and ROS-emitted by sustained oxidative phosphorylation are coupled during exercise recovery [99]. For an in-depth discussion of how mitophagy and mitochondrial turnover may regulate cellular bioenergetics for muscle remodeling, readers are referred to other recent publications [221,222].

It is important to note that the combined aerobic and mechanical stimulus induced by running-based models of EIMD make it difficult to determine whether autophagy is specifically induced to accommodate mechanically induced damage. In fact, many studies have confirmed that autophagic flux is upregulated in rodents following exercise without significant eccentric strain, *i.e.* uphill running and swimming [223–225], which is perhaps unsurprising considering autophagy's role in mitochondrial remodeling [226]. However, there is some evidence to suggest that eccentric loading promotes a unique autophagic response. Inducible ATG7-KO female mice display impaired exercise capacity compared to their autophagy-sufficient wild-type littermates during downhill, but not uphill, running [208], although limited morphological alterations were observed. In contrast, Lu et al. [227] showed that an unaccustomed bout of exhaustive wheel running induced immediate myofibrillar damage and elevated markers of chaperone-assisted selective autophagy (CASA) (see Tedesco et al. [228] for an in-depth CASA review). Considering skeletal muscle is more susceptible to damage from eccentric loading, an attractive hypothesis may be that resistance-type exercise induces a particularly potent autophagic response. However, in anesthetized rodents, electrically evoked eccentric, concentric, and isometric loading patterns all increase the phosphorylation of ULK1<sup>ser757</sup> and ULK1<sup>ser317</sup> to a similar degree without affecting LC3-II/LC3-I ratios [229]. Therefore, further investigations are warranted to determine whether autophagy flux is specifically increased in response to voluntary eccentric-EIMD.

## Autophagy may be upregulated in untrained human skeletal muscle recovering from novel resistance exercise

It is well known that RE, especially involving novel eccentric contractions [4,142], damages myofiber structure [6,136–139] and promotes a robust protein synthetic response [10,11,230]. In untrained individuals, RE-induced myofibrillar damage and protein synthesis are closely related, supporting the notion that muscle proteins are initially synthesized to renew damaged myofibers [5,6,231]. Despite this, human studies investigating the effect of unaccustomed RE on autophagy are equivocal (Table 1) [232–238].

One related hypothesis on this topic is that mechanical stimulation of AKT/mTORC1 activity inhibits autophagy following acute RE [18,235,239]. In support of this, Hentilä et al. [235] showed that LC3-II, but not LC3-I or p62, protein content decreased 1 hour post-exercise, in line with reductions in AMPK-dependent ULK1<sup>ser555</sup> and elevations in mTORC1-dependent ULK1<sup>ser757</sup> phosphorylation. Several autophagy-related proteins were, however, elevated once ULK1<sup>ser757</sup> returned to basal values 48 hours post exercise, which could suggest that autophagosome formation increases once anabolic signaling has diminished. Alternatively, it is possible that autophagy proteins may have accumulated due to inhibited autophagosome degradation during recovery, thereby corroborating the ‘bottleneck’ hypothesis [202]. Regulation of autophagy-related genes following novel RE is also equivocal, with studies reporting unaltered [233,235] or elevated [237] LC3 and p62 expression. The combination of novel RE and essential amino acid (EAA) ingestion, a further anabolic stimulus [8], elicits reductions in LC3-II/LC3-I ratio at 2 h post-exercise, possibly reflecting a reduction in autophagosome biogenesis. Interestingly, if the EAA beverage contained higher leucine doses, the LC3-II/LC3-I ratio increased at 5 and 24 hours, leading the authors to postulate that further elevations in mTORC1 activation may have impeded autophagosome degradation [238,240]. The observation that LC3-II increases with higher amounts of leucine ingestion is in contrast to *in vitro* [241] and animal investigations [242], which characterize leucine-mediated autophagy inhibition through a reduction in LC3-II content. However, as each of these studies only utilized ‘static’ assessments of autophagic protein content, more appropriate methodologies are required to better understand the relationship between RE, nutritional status, and autophagy flux.

Alike novel RE, Fritzen et al. [243] showed that 1 h one-legged concentric cycling exercise (80% peak workload with two 5-minute intervals at 100%) reduced LC3-II/LC3-I ratio, but not p62 content, for up to 4 hours post-exercise in moderately trained men. This occurred alongside elevated AMPK-dependent ULK1<sup>ser555</sup> phosphorylation, implying that while EE upregulates AMPK/ULK1 signaling, this may not lead to altered autophagosome content [243], or could simply reflect that static measurements are unable to accurately reflect autophagic flux. Other works have reported that exercise-induced ULK1<sup>ser555</sup> phosphorylation (AMPK-regulated), but not ULK1<sup>ser757</sup> (mTORC1), was associated with a lowered LC3-II/LC3-I ratio immediately after 1 h cycling at 50% maximal oxygen consumption (VO<sub>2max</sub>), potentially indicating elevated autophagosome clearance [244]. p62 protein content was, however, unaltered in this investigation, highlighting further uncertainty over whether autophagy flux was increased. In contrast, Mazo et al. [236] showed that acute cycling (40 minutes at 75% peak-heart rate) or lower-body RE reduced both LC3-II and p62 protein content to a similar degree in untrained individuals 4 hours after exercise. Considering that both LC3-II and p62 are degraded within the lysosomal lumen during autophagy [77], autophagosome degradation may have been upregulated, a notion reinforced by the observation that autophagy gene expression was upregulated at 1 h and 4 hours post-exercise. Nevertheless, while combined measures of p62, LC3-II, and upstream autophagic regulators provide a more detailed description of autophagy, these findings cannot confirm the status of autophagic flux, which requires the use of autophagosome-lysosome fusion inhibitors [77].

Despite the evident limitations of monitoring autophagy via static measures, Schwalm et al. [245] found that the LC3-II/LC3-I ratio and p62 protein content was decreased in endurance-trained individuals 1 hour after high-intensity (70% VO<sub>2max</sub>), but not low-intensity (55% VO<sub>2max</sub>) cycling. In addition, autophagy-related gene expression was greater in the high-intensity trial and these effects were primarily attributed to a greater induction of the AMPK/ULK1 axis, possibly indicating elevated autophagy induction and autophagosome clearance with higher intensity exercise. However, as the total amount of work performed differed between trials, it is unclear whether the decline in autophagy proteins was specifically related to exercise intensity. For example, work-matched bouts of exercise performed above, or below, maximum lactate steady state elicit similar reductions in LC3-II protein content albeit while p62 protein content and autophagy-related gene expression were unchanged [246]. These data may indicate that total work completed during an exercise bout may be the primary driver of alterations in autophagy, although further

**Table 1: Overview of human studies investigating effects of acute novel resistance exercise on skeletal muscle autophagy markers in healthy untrained individuals.**

Study	Participants	Exercise/Intervention	mRNA expression	Protein content
Díaz-Castro et al. (2024) (232)	Untrained adult males $n = 8$ Age $31.5 \pm 4.23$ y Weight $78.45 \pm 6.29$ kg	Unilateral leg presses 10 sets of 10 repetitions at 70% 1RM with 2 minute rest intervals	N/A	(vs. rested leg) LC3AB-II + 1 h↔ LC3AB-II/LC3AB-I + 1 h↔ p62 + 1 h↔ p-AMPK <sup>thr172</sup> + 1 h↔ p-mTOR <sup>ser448</sup> + 1 h↑ PINK1 + 1 h↔ Parkin + 1 h↓ BNIP3L/NIX + 1 h↓ FUNDC1 + 1 h↔ BNIP3 + 1 h↓ <sup>t2</sup> MFN2 + 1 h↓ <sup>t2</sup> OPA1 + 1 h↔ p-DRP1 <sup>ser616</sup> + 1 h↑
Dickinson et al. (2017) (238)	Untrained older males control group $n = 7$ Age $74 \pm 2$ y BMI $26 \pm 1$ Leucine group $n = 8$ Age $71 \pm 3$ y BMI $27 \pm 1$	Bilateral leg extensions 8 sets of 10 repetitions at 65% 1RM with 3-minute rest intervals 10g EEA beverages containing either 1.85 g leucine (control group) or 3.5 g leucine (leucine group) consumed 1 hour post exercise	Control group (vs. baseline) LC3 + 2 h↑ + 5 h↑ + 24 h↑ GABARAP + 2 h↔ + 5 h↔ + 24 h↔ BECN1 + 2 h↔ + 5 h↔ + 24 h↔ ATG7 + 2 h↔ + 5 h↔ + 24 h↔ LAMP2B + 2 h↔ + 5 h↔ + 24 h↔ CISD2 + 2 h↔ + 5 h↔ + 24 h↔ RUNX1 + 2 h↔ + 5 h↑ + 24 h↑ MuRF1 + 2 h↑ + 5 h↑ + 24 h↔ Leucine group (vs. baseline) LC3	Control group (vs. baseline) LC3B-I + 2 h↔ + 5 h↔ + 24 h↔ LC3B-II + 2 h↓ + 5 h↔ <sup>*1</sup> + 24 h↔ LC3B-II/LC3B-I + 2 h↓ + 5 h↓ <sup>*1</sup> + 24 h↓ <sup>*1</sup> Beclin-1 + 2 h↔ + 5 h↔ + 24 h↔ p-AKT <sup>thr308</sup> + 2 h↑ <sup>t2</sup> + 5 h↔ + 24 h↔ Nuclear/cytosolic FoxO3a + 2 h↔ + 5 h↔ + 24 h↔ <sup>*1</sup> Leucine group (vs. baseline) LC3B-I + 2 h↔ + 5 h↔ + 24 h↔ LC3B-II + 2 h↓ + 5 h↔ <sup>*1</sup> + 24 h↔

*Continued*

**Table 1: Continued.**

Study	Participants	Exercise/Intervention	mRNA expression	Protein content
Fry et al. (2013) (233)	Untrained younger adults <i>n</i> = 16 Age 27 ± 2 y BMI 25.1 ± 0.9	Bilateral leg extensions 8 sets of 10 repetitions at 70% 1RM with 3 minute rest intervals	+ 2 h↑ + 5 h↔ + 24 h↔	LC3B-II/LC3B-I
			GABARAP	+ 2 h↓ + 5 h↓* <sup>1</sup> + 24 h↔*Beclin-1 <sup>1</sup>
			+ 2 h↔ + 5 h↔ + 24 h↔	+ 2 h↔ + 5 h↔ + 24 h↔
			BECN1	p-AKT <sup>thr308</sup>
			+ 2 h↔ + 5 h↔ + 24 h↔	+ 2 h↑↑ + 5 h↔ + 24 h↔
			ATG7	Nuclear/cytosolic FoxO3a
			+ 2 h↔ + 5 h↔ + 24 h↔	+ 2 h↔ + 5 h↔ + 24 h↔* <sup>1</sup>
			LAMP2B	
			+ 2 h↔ + 5 h↔ + 24 h↔	
			CISD2	
			+ 2 h↔ + 5 h↔ + 24 h↑	
			RUNX1	
			+ 2 h↑↑ + 5 h↑ + 24 h↑	
			MuRF1	
			+ 2 h↑ + 5 h↑ + 24 h↔	
	Untrained older adults <i>n</i> = 16 Age 70 ± 2 y BMI 24.2 ± 0.6	Bilateral leg extensions 8 sets of 10 repetitions at 70% 1RM with 3 minute rest intervals	Younger group (vs. baseline)	Younger group (vs. baseline)
			LC3	LC3B-I
			+ 3 h↔ + 6 h↔ + 24 h↔	+ 3 h↔ + 6 h↔ + 24 h↔
			GABARAP	LC3B-II
			+ 3 h↓ + 6 h↓ <sup>†2</sup> + 24 h↔	+ 3 h↔ + 6 h↓ + 24 h↓
			MuRF1	LC3B-II/LC3B-I
			+ 3 h↑ + 6 h↑ + 24 h↔	+ 3 h↓ + 6 h↓ + 24 h↓
				Beclin-1
			Older group (vs. baseline)	+ 3 h↔* <sup>1</sup> + 6 h↔* <sup>1</sup> + 24 h↔* <sup>1</sup>
			LC3	ATG7
			+ 3 h↔ + 6 h↔ + 24 h↔	+ 3 h↔* <sup>1</sup> + 6 h↔* <sup>1</sup> + 24 h↑* <sup>1</sup>
			GABARAP	FoxO3a <sup>ser253</sup>
			+ 3 h↓ + 6 h↓ <sup>†2</sup> + 24 h↔	+ 3 h↓ + 6 h↓ + 24 h↓
			MuRF1	p-AKT <sup>thr308</sup>
			+ 3 h↑ + 6 h↑ + 24 h↔	+ 3 h↑ + 6 h↔ + 24 h↔
				Older group (vs. baseline)
				LC3B-I
				+ 3 h↔ + 6 h↔ + 24 h↔
				LC3B-II
				+ 3 h↓ + 6 h↓ + 24 h↓
				LC3B-II/LC3B-I
				+ 3 h↓ + 6 h↓ + 24 h↓
				Beclin-1
				+ 3 h↔* <sup>1</sup> + 6 h↔* <sup>1</sup> + 24 h↔* <sup>1</sup>
				ATG7
				+ 3 h↔* <sup>1</sup> + 6 h↔* <sup>1</sup> + 24 h↔* <sup>1</sup>
				FoxO3a <sup>ser253</sup>
				+ 3 h↓ + 6 h↓ + 24 h↓
				p-AKT <sup>thr308</sup>
				+ 3 h↑ + 6 h↔ + 24 h↔

*Continued*



**Table 1: Continued.**

Study	Participants	Exercise/Intervention	mRNA expression	Protein content
Glynn et al. (2010) (234)	Untrained adult males	Bilateral leg extensions 10 sets of 10	EAA + LCHO group (vs. baseline) MuRF1	EAA + LCHO group (vs. baseline) LC3B-I
	EAA + LCHO group <i>n</i> = 7	repetitions at 70%1RM with 3 minute rest intervals	+ 1 h↑ + 2 h↑	+ 1 h↔ + 2 h↔ LC3B-II
	Age 30 ± 2 y			+ 1 h↔ + 2 h↓
	BMI 26 ± 1	~20 g EAA beverage containing	EAA + HCHO group (vs. baseline) MuRF1	p-AMPK <sup>thr172</sup>
		0.35 g/kgLM EAA with either 0.5 g/ kgLM carbohydrate (EAA + LCHO group) or	+ 1 h↑ + 2 h↑	+ 1 h↑ + 2 h↑* <sup>1</sup> p-AKT <sup>ser473</sup>
	EAA + HCHO group <i>n</i> = 32 default 1	1.40 g/kgLM (EAA + HCHO group)		+ 1 h↑ + 2 h↑
	Age 32 ± 1 y	consumed straight after 1 hour biopsy		FoxO3a <sup>ser253</sup>
	BMI 27 ± 1	time point		+ 1 h↔ + 2 h↔ FoxO3a <sup>ser318/321</sup>
				+ 1 h↔ + 1 h↔ MuRF1
				+ 1 h↑ + 2 h↔
Hentilä et al. (2018) (235)	Untrained younger adult males <i>n</i> = 12 (whey protein group <i>n</i> = 6, placebo group <i>n</i> = 6)	Bilateral leg presses 5 sets of 10	Younger group (vs. baseline) LC3B	Younger group (vs. baseline) LC3B-I
	Age 27 ± 4 y	repetition maximums with 2 minute rest intervals	+ 1 h↔ + 48 h↔p62 + 1 h↔ + 48 h↔	+ 1 h↔ + 48 h↑ LC3B-II
				+ 1 h↓ + 48 h↑p62
	Untrained older adults <i>n</i> = 8	Young cohort consumed either 15 g whey protein isolate or an isocaloric placebo immediately before and after the resistance exercise bout (pooled analysis, no effect of supplementation on autophagy markers)		+ 1 h↔ + 48 h↑ p-ULK1 <sup>ser555</sup>
	Age 61 ± 6 y			+ 1 h↓ + 48 h↔ p-ULK1 <sup>ser757</sup>
	BMI 23.4–28.8			+ 1 h↑↑ + 48 h↔ Beclin-1
				+ 1 h + 48 h↑↑ <sup>2</sup> BCL-2
				+ 1 h↔ + 48 h↔

*Continued*

**Table 1: Continued.**

Study	Participants	Exercise/Intervention	mRNA expression	Protein content
				Older group (vs. baseline)
				LC3B-I
				+ 48 h↔
				LC3B-II
				+ 48 h↔
				p62 + 48 h↔
				Beclin-1
				+ 48 h↔BCL-2
				+ 48 h↔
Mazo et al. (2021) (236)	Untrained adult males <i>n</i> = 6 Age 27 ± 3 y Weight 79 ± 10 kg	Unilateral leg extensions 8 sets of 10 repetitions at 60–65%1RM with 3 minute rest intervals	Resistance exercise (vs. baseline) Autophagy genes + 1 h 7 ATGs DE + 4 h 43 ATGsDE FOXO3 pathway + 1 h↑ + 4 h↑ mTOR pathway + 1 h? + 4 h↑	Resistance exercise (vs. baseline) LC3B-I + 1 h↔ + 4 h↔ LC3B-II + 1 h↓ + 4 h↓ LC3B-II/LC3B-I + 1 h↔ + 4 h↔ p62 + 1 h↔ + 4 h↓ FoxO3a + 1 h↔ + 4 h↓ mTOR <sup>ser2448</sup> + 1 h↔ + 4 h↑*1
		40 minutes of stationary cycling at 75% peak heart rate		
		Cross-over trial comparing an unaccustomed bout of aerobic and resistance exercise, separated by ~ 1 week	Aerobic exercise (vs. baseline) Autophagy genes + 1 h 6 ATGs DE + 4 h 17 ATGsDE FOXO3 pathway + 1 h↑ + 4 h? mTOR pathway + 1 h? + 4 h↑	Aerobic exercise (vs. baseline) LC3B-I + 1 h↔ + 4 h↔ LC3B-II + 1 h↓ + 4 h↓ LC3B-II/LC3B-I + 1 h↔ + 4 h↔ P62 + 1 h↔ + 4 h↓ FoxO3a + 1 h↔ + 4 h↓ mTOR <sup>ser2448</sup> + 1 h↔ + 4 h↑*21
Ogborn et al. (2015) (237)	Untrained younger males <i>n</i> = 9 Age 21 ± 3 y Weight 91.7 ± 21.9 kg Untrained older males <i>n</i> = 9 Age 70 ± 4 Weight 87.6 ± 11.5 kg	Unilateral leg extensions and leg presses 4 sets of 10 repetitions per exercise at 75%1RM with 2-minute rest intervals (pooled analysis, no effect of age)	(vs. rested leg) LC3B + 3 h↑ + 24 h↑ <sup>†2</sup> + 48 h↔ p62 + 3 h↑ + 24 h↔ + 48 h↔ ATG7 + 3 h↔ + 24 h↔ + 48 h↔ BECN1 + 3 h↔ + 24 h↔ + 48 h↔	(vs. rested leg) Total LC3B + 3 h↔ + 24 h↑ <sup>†2</sup> + 48 h↑ p62 + 3 h↔ + 24 h↑ + 48 h↑ ATG7 + 3 h↔ + 24 h↔ + 48 h↔ PINK1 + 3 h↔ + 24 h↔

*Continued*

**Table 1: Continued.**

Study	Participants	Exercise/Intervention	mRNA expression	Protein content
			VPS34	+ 48 h↔
			+ 3 h↔ + 24 h↔ + 48 h↔	Parkin
			BNIP3	+ 3 h↔ + 24 h↔ + 48 h↔
			+3h↔+24h↔ + 48 h↔	

<sup>1</sup>Denotes a significant difference between condition/group.

<sup>2</sup>Indicates a borderline significant difference compared to baseline or rested sample.

work utilizing more sophisticated measures of autophagic flux in human skeletal muscle is required to confirm this.

In addition to EE, RT can induce mitochondrial remodeling [247–251]; thus, it is plausible that auto/mitophagy pathways may be regulated by this form of exercise. Diaz-Castro et al. [232] recently reported that acute RE increased markers of mitochondrial fission and elicited reductions in the protein content of the mitophagy receptor BNIP3L/NIX in untrained human muscle. TEM-derived observations of damaged mitochondria and mitophagosome-like structures were also noted at this time, suggesting that mitophagy may have been taking place. It is unclear whether the mitophagosome-like structures were degraded intracellularly as LC3-II and p62 content were unchanged. Intriguingly, though, it was also reported that subsarcolemmal mitophagosome-like structures could be seen exiting muscle following RE, a phenomenon that has been described in other cell types [252,253]. While Diaz-Castro et al. [232] did not show evidence of mitophagosomes in circulation, other studies have shown that mouse cardiac muscle with impaired lysosomal function also ejects damaged mitochondria, but the vesicles do not appear in circulation due to being degraded by nearby macrophages [254]. Similar ‘outsourcing’ of mito-/autophagy has also been observed within human mesenchymal stem cells [255], and pharmaceutical inhibition of lysosomal function elevates secretory autophagy [256,257]. Considering these data, extracellular release of autophagosome-like structures could represent an alternative mechanism to eliminate cellular debris when lysosomes are inundated during recovery from novel RE (i.e. the ‘bottleneck’) [232], although further research regarding this novel hypothesis is required.

## Limitations of this field and outstanding questions

### Static measures of autophagy are insufficient to determine autophagic flux

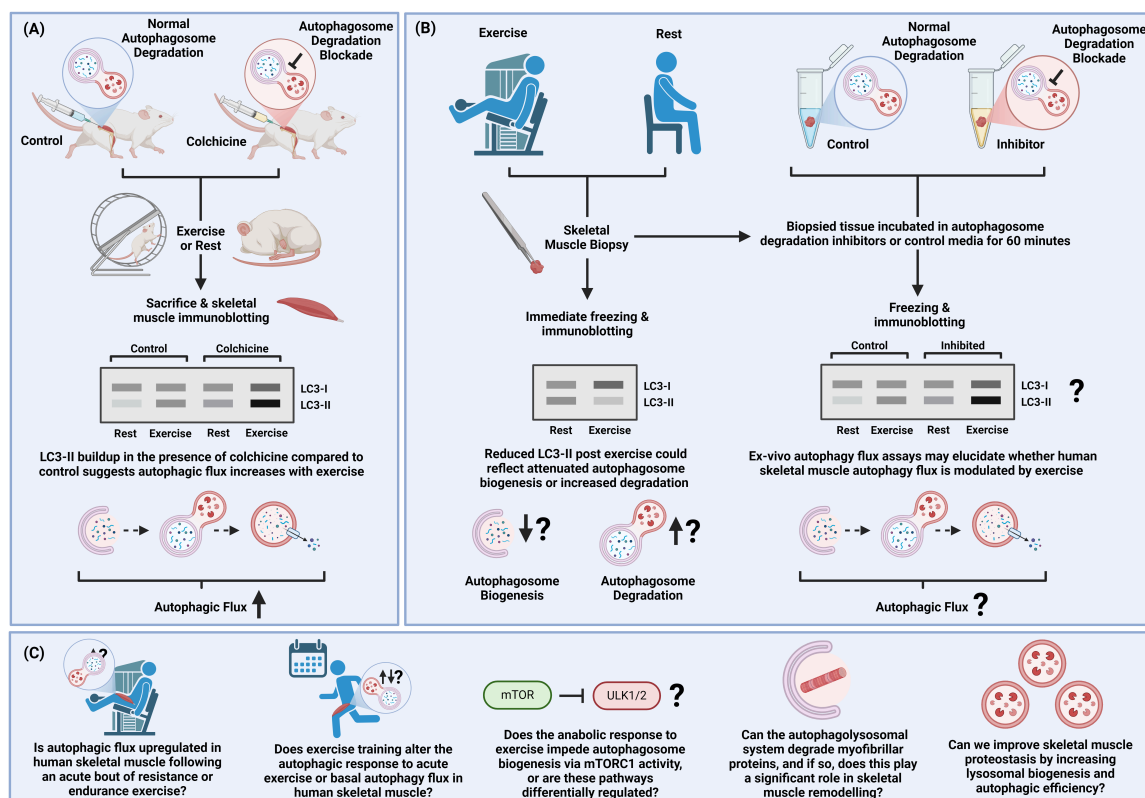
As emphasized throughout this review, a key limitation of human research is the use of static markers to infer the status of autophagy flux. Cellular contents of LC3, p62, and many other autophagy-related proteins change depending on their rate of synthesis and/or their autophagic degradation, meaning that unidirectional changes could be due to either elevated autophagosome formation or reduced fusion with lysosomes and subsequent degradation. Drugs that block autophagosome degradation (e.g. colchicine and bafilomycin) have proven invaluable in determining the autophagic effect of exercise in animals and cellular models of muscle contraction [77]; however, the use of these drugs in human research is ethically challenging. By adapting such an assay [258], Botella et al. [246] have recently provided some, albeit limited, evidence that EE may upregulate autophagy flux in human skeletal muscle. In agreement with most investigations, static measures of human LC3-II protein content were immediately reduced post-exercise and returned to basal values within 3.5 hours of recovery, with no changes in p62 protein content or related mRNA expression. In contrast, in rodents, LC3-II was unaffected immediately after but increased 3.5 hours post-exercise, corroborating most reports that static measures of LC3-II decrease in human [233–236,238,243–245] and increase in rodent [17,110,111,208,217,227,259–261] skeletal muscle during the initial stages of exercise recovery. However, when a small subset of biopsied tissue ( $n = 5$ ) was incubated in a lysosomotropic ammonium chloride-leupeptin solution prior to freezing, a moderate-to-large effect of exercise on elevating LC3-II flux was observed for up to 24 hours [246]. These data should be interpreted with caution given the limited sample size, but they do indicate the potential for this methodology to be utilized in human exercise studies.

Overall, comparing human and rodent changes in static measures of LC3 may lead to false conclusions regarding the autophagic response to exercise. Although a confirmatory lysosomal blockade can delineate

whether LC3-II is altered by lysosomal degradation, LC3-II is also a feature of phagocytic and endocytic pathways that converge at the lysosome [262]. Therefore, the flux of other autophagy-related proteins could also be included to provide a better indication of whether the autophagolysosomal system is specifically upregulated. We hope that future experiments employing *ex vivo* autophagy assays will shed light on conflicting data and ultimately begin to decipher the complexity of autophagy regulation in human skeletal muscle (Figure 4).

## Potential mTOR-independent mechanisms of selective-autophagy at sites of cellular damage

A primary reason why many researchers postulate that RE could lead to inhibition of autophagy is due to mTORC1's inhibitory effects on aspects of the autophagic cellular machinery [78,86]. However, it is becoming increasingly evident that autophagic pathways can be differentially regulated [263,264], and several accounts of mTORC1-independent autophagic induction have been described in *in vitro* and animal studies. For example, Cardenas et al. [265,266] identified an AMPK-mediated mechanism of autophagy activation in response to alterations in ER-mitochondria calcium flux, which occurs regardless of mTORC1 activity status. Moreover, others have displayed ULK1-mediated autophagy to occur at mitochondria and peroxisomes independent of both AMPK and mTORC1 [267] and investigations in C2C12 myotubes show that CASA-mediated degradation of filamin can occur despite mechanically induced mTORC1 activation [268]. In addition to autophagy induction events, transcriptional regulation of the autophagolysosomal system can occur independent of mTORC1. Medina et al. [108] reported that local calcium release from the lysosomal calcium channel MCOLN1 activates nearby calcineurin, which, in turn, dephosphorylates TFEB<sup>Ser211/Ser142</sup> allowing its nuclear translocation even in nutrient-replete conditions.



**Figure 4: Limitations of current research and future questions.**

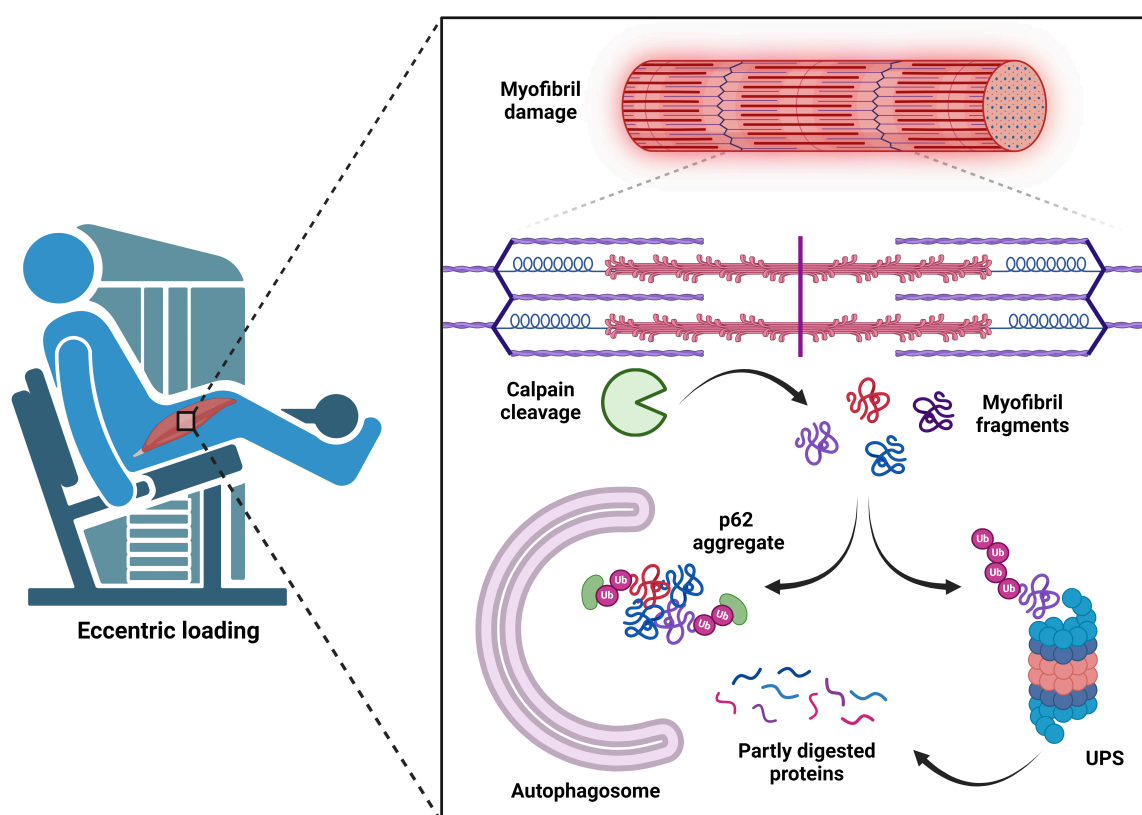
(A) Experimental model commonly used to measure autophagic flux in rodents. (B) Schematic of how an *ex vivo* autophagy flux methodology could be incorporated into existing human muscle biopsy techniques to better infer activity of the autophagolysosomal system. (C) A list of pertinent questions yet to be elucidated by researchers studying human skeletal muscle autophagy.



The precise mechanism governing MCOL1N activation in exercised skeletal muscle is unclear, although mitochondrial-ROS have been shown to stimulate the MCOL1N/TFEB axis in COS-1 and HEK293 cells [106,107]. It is plausible that such mTORC1-independent mechanisms could occur in human skeletal muscle following RE to elevate autophagic flux, albeit well-controlled experimental investigations are required to elucidate this.

## Does autophagy contribute to myofibrillar protein degradation?

Another issue often discussed in the field of muscle protein metabolism is whether autophagy can contribute to myofibrillar protein turnover. Early research demonstrated that lysosomal protease inhibitors do not suppress proxy measures of myofibrillar protein breakdown [269], and immunofluorescence imaging of starved rat skeletal muscle shows cytoplasmic autophagosomes with no evidence of enclosed myofibrillar proteins [25]. Therefore, it is generally believed that the proteasomal and calpain systems are primarily responsible for myofibrillar protein turnover while the lysosomal system facilitates degradation of cytosolic proteins and organelles [20,270]. However, *in vitro* investigations have shown that lysosomal cathepsins can hydrolyze purified myofibrillar proteins [271–273] and recent transmission electron microscopy (TEM) images of human and rodent skeletal muscle following EIMD observe autophagosome-like structures within the intermyofibrillar space [127,207,227,274]. Furthermore, autophagic flux is upregulated in aging rodent skeletal muscle with impaired proteasomal function and significant protein aggregation, possibly reflecting compensation of the autophagy system [18]. Interestingly, autophagic flux is not further enhanced in these animals during recovery from disuse atrophy, which could suggest that lysosomes are inundated in basal conditions and cannot cope with an elevated need to degrade accumulating aggregates. Therefore, autophagy may operate in harmony with other proteolytic pathways to



**Figure 5: Hypothetical model of contractile protein degradation by the autophagy system.**

Myofibrillar proteins can become damaged during periods of mechanical loading and must, therefore, be removed for muscle recovery to ensue. The calpains release myofibrillar proteins from sarcomeres allowing the ubiquitinated fragments to be degraded by the UPS. The autophagy system may also contribute to the degradation of aggregated myofibrillar fragments or partly degraded peptides released from the UPS. Created in BioRender. Acheson, J. (2024) BioRender.com/f57e974.

sufficiently degrade cleaved contractile proteins or aggregated peptide chains during periods of myofibrillar damage (Figure 5).

One such investigation that has observed autophagy-related myofibrillar degradation in humans is Ulbricht et al. [274], whereby an acute bout of maximal eccentric contractions, but not conventional RE, elicited myofibrillar disruption and reductions in CASA proteins and their substrate, filamin C. Immunofluorescence imaging of damaged fibers showed an increase in LC3-positive structures suggesting autophagosome presence/formation in this region. Furthermore, the mechanical stretch of the myofibrillar protein titin in rat cardiomyocytes exposes a cryptic titin-kinase binding site, which can associate with autophagy adaptors neighbor of BRCA1 gene 1 (NBR1) and p62 [275]. More recent data indicate that muscle inactivity may promote an interaction between NBR1/p62 and titin-kinase at sarcomeres, further indicating a potential role of autophagy in myofibrillar turnover [276]. Nevertheless, further work is required to confirm whether autophagy flux is enhanced during periods where CASA protein content decreases [274] as well as to determine whether this pathway contributes to myofibrillar protein turnover.

## Is autophagy involved in skeletal muscle adaptation to chronic exercise?

The repeated bout effect increases skeletal muscle's resilience to mechanical damage [4], contributed to by a variety of adaptations including a sensitized immune response, extracellular matrix remodeling, and neural modulation [3]. However, there is some evidence that the autophagy system is also modulated by consistent training. Ulbricht et al. [274] observed protein levels of p62 and several CASA components to be elevated after 4 weeks of progressive RT, but not constant load RT, suggesting that progressive overload is required for adaptation to the autophagolysosomal system. Conversely, others have shown no changes in p62 protein content following 12 weeks of progressive RT [277], although this study was conducted in trained men where adaptations could have already occurred. It is also possible that different types of RE may induce unique adaptations in the autophagy system. Lim et al. [278] reported that 10 weeks of low-load, high-volume RT (30%1RM) performed to volitional fatigue increased Parkin protein content and proteins involved in mitochondrial dynamics, yet these adaptations did not occur in high-load or low-intensity non-failure training cohorts. Thus, high-volume, low-load RT may promote a greater capacity for mitochondrial remodeling, possibly to accommodate mitochondrial stress elicited by a more 'endurance-type' exercise. Indeed, 8 weeks of continuous moderate-intensity or work-matched sprint interval endurance training (ET) increases Parkin, BNIP3, LC3-I, and oxidative phosphorylation complex 1 content in previously moderately trained men [279]. Similarly, in mouse skeletal muscle, 6 weeks of ET increased mitochondria content, Parkin expression, and Parkin-colocalization with mitochondrial markers [280], although basal levels of mitophagy flux were unchanged, further highlighting the importance of including measurements of flux in such investigations. Other studies investigating the effects of chronic exercise training on 'static' measures of autophagy protein content observe contrasting results with several reporting increased LC3-I content [243,279], which could indicate increased autophagic capacity without alterations in basal autophagosome content, while others suggest that the LC3-II content is increased, indicating potential expansion of the autophagosome pool [235,246].

Another model that has provided insight into the regulation of the autophagolysosomal system to exercise training is that of chronic contractile activity (CCA), employed consistently by the laboratory of Prof. David Hood. Here, several investigations have indicated that frequent bouts of muscle contraction enhance the content of proteins that regulate autophagy induction (e.g. LC3-I, Beclin-1) and related transcriptional programs [100,281–283]. Paradoxically, however, when utilizing autophagy inhibitors, CCA had either no effect or reduced basal LC3-II/p62 flux, potentially reflecting improvements in muscle quality [100,281,283,284]. The acute increase in autophagy flux during EE recovery is also attenuated in trained-mouse quadriceps, indicating increased resilience to exercise-induced stress [281]. Similarly, 9 weeks of progressive weighted climbing exercise reduces markers of autophagy flux (e.g. LC3-II/LC3-I ratio and p62) while increasing those of autophagic and lysosomal capacity in aging rats [285]. *In vitro* data also support these notions as CCA elicits elevations in TFEB protein content and markers of lysosomal content and proteolytic activity [100,282,284]. Notably, evidence of lysosomal biogenesis can occur within as little as 3 days of increased contractile activity and precedes mitochondrial adaptations in these models [282]. In contrast, TFEB content was unaltered in young mouse skeletal muscle following 9 days of CCA *in vivo* [283], although lysosomal biogenesis did occur. Overall, these data show that TFEB activity and lysosome

biogenesis are important mechanisms underpinning skeletal muscle plasticity and may contribute to other autophagic adaptations.

It is unclear whether TFEB nuclear translocation and lysosomal biogenesis are enhanced by exercise training in human skeletal muscle, although we have observed elevations in LAMP2 protein content following 8 weeks of progressive RE [286]. Considering the relationship between CCA and lysosomal biogenesis *in vitro* and in rodent skeletal muscle, we speculate that an enhanced lysosomal pool may increase the efficiency of autophagolysosomal recycling [281]. Furthermore, given that lysosomal biogenesis appears to be the rate-limiting factor of the autophagolysosomal system during skeletal muscle repair (i.e. the 'bottleneck' [202]), methods to improve lysosomal capacity may benefit untrained individuals susceptible to EIMD. *In vitro* and animal-based studies have reported that nutraceutical compounds such as curcumin [282,283], spermidine [284,285,287], and quercetin [288,289] can promote lysosomal biogenesis and autophagic function, although the use of these in relation to exercise-induced autophagy have yet to be comprehensively investigated in human skeletal muscle. Importantly, some evidence suggests that antioxidant supplementation can also attenuate autophagy [103,208], highlighting the need for further mechanistic investigation of these compounds.

## Conclusion

The autophagolysosomal system maintains skeletal muscle homeostasis throughout the lifespan and during acute stress such as energy imbalance and tissue injury. Changes in intracellular calcium and redox status are key signals regulating autophagy induction and transcriptional programs. Animal models generally indicate that autophagy is upregulated to remove dysfunctional mitochondria and preserve skeletal muscle integrity during recovery from strenuous exercise. However, a lack of sufficient methods to monitor autophagy flux has prevented any robust conclusions regarding whether autophagic degradation is increased by exercise-related stress in human skeletal muscle. It is well established that novel eccentric exercise is particularly damaging to myofibrillar protein architecture; thus, autophagy may play an important role in muscle regeneration and remodeling. Moreover, consistent exercise training can increase the expression of autophagy-related genes and proteins, suggesting potential adaptation of the autophagy system to increased skeletal muscle loading. Nonetheless, further well-controlled human investigations utilizing various autophagic signaling and flux measurements are required to appropriately delineate whether, and if so, how autophagy contributes to exercise-induced muscle repair/remodeling.

## Conflicts of Interest

The authors declare that they have no conflicts of interest.

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## CRedit Author Contribution

J.A. drafted manuscript; J.A. prepared figures; J.A., S.J., C.S., and N.H. edited and revised manuscript;

J.A., S.J., C.S., and N.H. approved final version of manuscript.

## Abbreviations

AKT, protein kinase B/AKT; AMP, adenosine monophosphate; AMPK, 5' AMP-activated protein kinase; ATG, autophagy-related gene; ATP, adenosine triphosphate; BCL-2, B-cell leukemia/lymphoma 2 protein; BNIP3, BCL2/Adenovirus E1B 19 kDa protein-interacting protein 3; BNIP3L/NIX, BCL2/Adenovirus E1B 19 kDa protein-interacting protein 3-like; CASA, chaperone-assisted selective autophagy; CCA, chronic contractile activity; CLEAR, coordinated lysosomal enhancement and regulation; CaMKK $\beta$ , calcium/calmodulin-dependent protein kinase kinase  $\beta$ ; Ctx, cardio-toxin; EAA, essential amino acids; EDL, extensor digitorum longus; EE, endurance exercise; EIMD, exercise-induced muscle damage; ER, endoplasmic reticulum; ERK1/2, extracellular signal-regulated kinases; ESCRT, endosomal sorting complexes required for transport; ET, endurance training; FIP200, focal adhesion kinase family interaction protein of 200kD; FOXO3, forkhead box O3; GABARAP, gamma-aminobutyric acid receptor-associated protein; HMGB1, high mobility group box-1 protein; HOPS, homotypic

fusion and protein sorting; I/R, ischemia reperfusion; JNK1, c-Jun N-terminal protein kinase 1; KO, knockout; LAMP2, lysosome-associated membrane protein 2; LC3, microtubule-associated protein 1A/1B-light chain 3; MAPK, mitogen-activated protein kinases; MCOLN1, mucolipin-1/TRPML1; MPB, muscle protein breakdown; MPS, muscle protein synthesis; MiT/TFE, microphthalmia/transcription factor E; NBR1, neighbor of BRCA1 gene 1; PE, phosphatidylethanolamine; PI3K-C1, class III phosphatidylinositol 3-kinase complex 1; PINK1, PTEN-induced putative protein kinase 1; PI3P, phosphatidylinositol 3-phosphate; PIS, phosphatidylinositol synthase; PKC $\theta$ , protein kinase C theta; RE, resistance exercise; ROS, reactive oxygen species; RT, resistance training; SNARE, soluble N-ethylmaleimide-sensitive factor attachment protein receptors; SR, sarcoplasmic reticulum; TEM, transmission electron microscopy; TFE3, transcription Factor Binding to IGHM Enhancer 3; TFE3, transcription factor EB; ULK1/2, unc51-like kinase 1/2; UPS, ubiquitin-proteasome; VAPs, VAMP-associated proteins; VO<sub>2max</sub>, maximal oxygen consumption; VPS, vacuolar protein sorting; WIPI, WD repeat domain; mTORC1, mammalian target of rapamycin complex 1; p62, sequestosome-1; v-ATPase, vacuolar H<sup>+</sup>-adenosine triphosphatases.

## References

- Paulsen, G., Mikkelsen, U.R., Raastad, T. and Peake, J.M. (2012) Leucocytes, cytokines and satellite cells: what role do they play in muscle damage and regeneration following eccentric exercise? *Exerc. Immunol. Rev.* **18**, 42–97
- Owens, D.J., Twist, C., Cobley, J.N., Howatson, G. and Close, G.L. (2019) Exercise-induced muscle damage: what is it, what causes it and what are the nutritional solutions? *Eur. J. Sport Sci.* **19**, 71–85 <https://doi.org/10.1080/17461391.2018.1505957>
- Hylidahl, R.D., Chen, T.C. and Nosaka, K. (2017) Mechanisms and mediators of the skeletal muscle repeated bout effect. *Exerc. Sport Sci. Rev.* **45**, 24–33 <https://doi.org/10.1249/JES.0000000000000095>
- Stupka, N., Tarnopolsky, M.A., Yardley, N.J. and Phillips, S.M. (2001) Cellular adaptation to repeated eccentric exercise-induced muscle damage. *J. Appl. Physiol.* **91**, 1669–1678 <https://doi.org/10.1152/jappl.2001.91.4.1669>
- Damas, F., Libardi, C.A. and Ugrinowitsch, C. (2018) The development of skeletal muscle hypertrophy through resistance training: the role of muscle damage and muscle protein synthesis. *Eur. J. Appl. Physiol.* **118**, 485–500 <https://doi.org/10.1007/s00421-017-3792-9>
- Damas, F., Phillips, S.M., Libardi, C.A., Vechin, F.C., Lixandrão, M.E., Jannig, P.R. et al. (2016) Resistance training-induced changes in integrated myofibrillar protein synthesis are related to hypertrophy only after attenuation of muscle damage. *J. Physiol. (Lond.)* **594**, 5209–5222 <https://doi.org/10.1113/JP272472>
- Green, H., Goreham, C., Ouyang, J., Ball-Burnett, M. and Ranney, D. (1999) Regulation of fiber size, oxidative potential, and capillarization in human muscle by resistance exercise. *Am. J. Physiol. Regul. Integr. Comp. Physiol.* **276**, R591–R596 <https://doi.org/10.1152/ajpregu.1999.276.2.R591>
- Kumar, V., Atherton, P., Smith, K. and Rennie, M.J. (2009) Human muscle protein synthesis and breakdown during and after exercise. *J. Appl. Physiol.* **106**, 2026–2039 <https://doi.org/10.1152/japplphysiol.91481.2008>
- Phillips, S.M. (2014) A brief review of critical processes in exercise-induced muscular hypertrophy. *Sports Med.* **44** Suppl 1, S71–7 <https://doi.org/10.1007/s40279-014-0152-3>
- Moore, D.R., Phillips, S.M., Babraj, J.A., Smith, K. and Rennie, M.J. (2005) Myofibrillar and collagen protein synthesis in human skeletal muscle in young men after maximal shortening and lengthening contractions. *Am. J. Physiol. Endocrinol. Metab.* **288**, E1153–9 <https://doi.org/10.1152/ajpendo.00387.2004>
- Phillips, S.M., Parise, G., Roy, B.D., Tipton, K.D., Wolfe, R.R. and Tarnopolsky, M.A. (2002) Resistance-training-induced adaptations in skeletal muscle protein turnover in the fed state. *Can. J. Physiol. Pharmacol.* **80**, 1045–1053 <https://doi.org/10.1139/y02-134>
- Phillips, S.M., Tipton, K.D., Ferrando, A.A. and Wolfe, R.R. (1999) Resistance training reduces the acute exercise-induced increase in muscle protein turnover. *Am. J. Physiol.* **276**, E118–24 <https://doi.org/10.1152/ajpendo.1999.276.1.E118>
- Nosaka, K., Lavender, A., Newton, M. and Sacco, P. (2003) Muscle damage in resistance training - is muscle damage necessary for strength gain and muscle hypertrophy. *Int. J. Sport Health Sci.* **1**, 1–8 <https://doi.org/10.5432/ijshs.1.1>
- Schoenfeld, B.J. (2012) Does exercise-induced muscle damage play a role in skeletal muscle hypertrophy? *J. Strength Cond. Res.* **26**, 1441–1453 <https://doi.org/10.1519/JSC.0b013e31824f207e>
- Pavis, G.F., Jameson, T.S.O., Dirks, M.L., Lee, B.P., Abdelrahman, D.R., Murton, A.J. et al. (2021) Improved recovery from skeletal muscle damage is largely unexplained by myofibrillar protein synthesis or inflammatory and regenerative gene expression pathways. *Am. J. Physiol. Endocrinol. Metab.* **320**, E291–E305 <https://doi.org/10.1152/ajpendo.00454.2020>
- Fuqua, J.D., Lawrence, M.M., Hettinger, Z.R., Borowik, A.K., Brecheen, P.L., Szczygiel, M.M. et al. (2023) Impaired proteostatic mechanisms other than decreased protein synthesis limit old skeletal muscle recovery after disuse atrophy. *J. Cachexia. Sarcopenia. Muscle.* **14**, 2076–2089 <https://doi.org/10.1002/jcsm.13285>
- Abbott, C.B., Lawrence, M.M., Kobak, K.A., Lopes, E.B.P., Peelor, F.F., 3rd, Donald, E.J. et al. (2021) A novel stable isotope approach demonstrates surprising degree of age-related decline in skeletal muscle collagen proteostasis. *Funct. (Oxf.)* **2**, zqab028 <https://doi.org/10.1093/function/zqab028>
- Baehr, L.M., West, D.W.D., Marcotte, G., Marshall, A.G., De Sousa, L.G., Baar, K. et al. (2016) Age-related deficits in skeletal muscle recovery following disuse are associated with neuromuscular junction instability and ER stress, not impaired protein synthesis. *Aging (Milano)* **8**, 127–146 <https://doi.org/10.18632/aging.100879>
- Tipton, K.D., Hamilton, D.L. and Gallagher, I.J. (2018) Assessing the role of muscle protein breakdown in response to nutrition and exercise in humans. *Sports Med.* **48**, 53–64 <https://doi.org/10.1007/s40279-017-0845-5>
- Goll, D.E., Neti, G., Mares, S.W. and Thompson, V.F. (2008) Myofibrillar protein turnover: the proteasome and the calpains. *J. Anim. Sci.* **86**, E19–35 <https://doi.org/10.2527/jas.2007-0395>



- 21 De Duve, C. and Wattiaux, R. (1966) Functions of lysosomes. *Annu. Rev. Physiol.* **28**, 435–492 <https://doi.org/10.1146/annurev.ph.28.030166.002251>
- 22 Ohsumi, Y. (1999) Molecular mechanism of autophagy in yeast, *Saccharomyces cerevisiae*. *Philos. Trans. R. Soc. Lond., B, Biol. Sci.* **354**, 1577–1580 <https://doi.org/10.1098/rstb.1999.0501>
- 23 Tsukada, M. and Ohsumi, Y. (1993) Isolation and characterization of autophagy-defective mutants of *Saccharomyces cerevisiae*. *FEBS Lett.* **333**, 169–174 [https://doi.org/10.1016/0014-5793\(93\)80398-e](https://doi.org/10.1016/0014-5793(93)80398-e)
- 24 Masiero, E., Agatea, L., Mammucari, C., Blaauw, B., Loro, E., Komatsu, M. et al. (2009) Autophagy is required to maintain muscle mass. *Cell Metab.* **10**, 507–515 <https://doi.org/10.1016/j.cmet.2009.10.008>
- 25 Mizushima, N., Yamamoto, A., Matsui, M., Yoshimori, T. and Ohsumi, Y. (2004) In vivo analysis of autophagy in response to nutrient starvation using transgenic mice expressing a fluorescent autophagosome marker. *Mol. Biol. Cell.* **15**, 1101–1111 <https://doi.org/10.1091/mbc.e03-09-0704>
- 26 Call, J.A., Wilson, R.J., Laker, R.C., Zhang, M., Kundu, M. and Yan, Z. (2017) Ulk1-mediated autophagy plays an essential role in mitochondrial remodeling and functional regeneration of skeletal muscle. *Am. J. Physiol., Cell Physiol.* **312**, C724–C732 <https://doi.org/10.1152/ajpcell.00348.2016>
- 27 Franco-Romero, A. and Sandri, M. (2021) Role of autophagy in muscle disease. *Mol. Aspects Med.* **82**, 101041 <https://doi.org/10.1016/j.mam.2021.101041>
- 28 Wang, C., Wang, H., Zhang, D., Luo, W., Liu, R., Xu, D. et al. (2018) Phosphorylation of ULK1 affects autophagosome fusion and links chaperone-mediated autophagy to macroautophagy. *Nat. Commun.* **9**, 3492 <https://doi.org/10.1038/s41467-018-05449-1>
- 29 Kaushik, S. and Cuervo, A.M. (2018) The coming of age of chaperone-mediated autophagy. *Nat. Rev. Mol. Cell Biol.* **19**, 365–381 <https://doi.org/10.1038/s41580-018-0001-6>
- 30 Wang, L., Klionsky, D.J. and Shen, H.M. (2023) The emerging mechanisms and functions of microautophagy. *Nat. Rev. Mol. Cell Biol.* **24**, 186–203 <https://doi.org/10.1038/s41580-022-00529-z>
- 31 Kirkin, V. and Rogov, V.V. (2019) A diversity of selective autophagy receptors determines the specificity of the autophagy pathway. *Mol. Cell.* **76**, 268–285 <https://doi.org/10.1016/j.molcel.2019.09.005>
- 32 Karanasios, E., Stapleton, E., Manifava, M., Kaizuka, T., Mizushima, N., Walker, S.A. et al. (2013) Dynamic association of the ULK1 complex with omegasomes during autophagy induction. *J. Cell. Sci.* **126**, 5224–5238 <https://doi.org/10.1242/jcs.132415>
- 33 Broadbent, D.G., Barnaba, C., Perez, G.I. and Schmidt, J.C. (2023) Quantitative analysis of autophagy reveals the role of ATG9 and ATG2 in autophagosome formation. *J. Cell Biol.* **222**, e202210078 <https://doi.org/10.1083/jcb.202210078>
- 34 Fracchiolla, D., Chang, C., Hurley, J.H. and Martens, S. (2020) A PI3K-WIP2 positive feedback loop allosterically activates LC3 lipidation in autophagy. *J. Cell Biol.* **219**, e201912098 <https://doi.org/10.1083/jcb.201912098>
- 35 Axe, E.L., Walker, S.A., Manifava, M., Chandra, P., Roderick, H.L., Habermann, A. et al. (2008) Autophagosome formation from membrane compartments enriched in phosphatidylinositol 3-phosphate and dynamically connected to the endoplasmic reticulum. *J. Cell Biol.* **182**, 685–701 <https://doi.org/10.1083/jcb.200803137>
- 36 Zhao, Y.G., Liu, N., Miao, G., Chen, Y., Zhao, H. and Zhang, H. (2018) The ER contact proteins VAPA/B interact with multiple autophagy proteins to modulate autophagosome biogenesis. *Curr. Biol.* **28**, 1234–1245 <https://doi.org/10.1016/j.cub.2018.03.002>
- 37 van Vliet A.R., Chidzuza, G.N., Maslen, S.L., Pye, V.E., Joshi, D., De Tito, S. et al. (2022) ATG9A and ATG2A form a heteromeric complex essential for autophagosome formation. *Mol. Cell.* **82**, 4324–4339 <https://doi.org/10.1016/j.molcel.2022.10.017>
- 38 Maeda, S., Otomo, C. and Otomo, T. (2019) The autophagic membrane tether ATG2A transfers lipids between membranes. *Elife* **8**, e45777 <https://doi.org/10.7554/eLife.45777>
- 39 Kannangara, A.R., Poole, D.M., McEwan, C.M., Youngs, J.C., Weerasekara, V.K., Thornock, A.M. et al. (2021) BiolD reveals an ATG9A interaction with ATG13-ATG101 in the degradation of p62/SQSTM1-ubiquitin clusters. *EMBO Rep.* **22**, e51136 <https://doi.org/10.15252/embr.202051136>
- 40 Young, A.R.J., Chan, E.Y.W., Hu, X.W., Köchl, R., Crawshaw, S.G., High, S. et al. (2006) Starvation and ULK1-dependent cycling of mammalian Atg9 between the TGN and endosomes. *J. Cell. Sci.* **119**, 3888–3900 <https://doi.org/10.1242/jcs.03172>
- 41 Olivas, T.J., Wu, Y., Yu, S., Luan, L., Choi, P., Guinn, E.D. et al. (2023) ATG9 vesicles comprise the seed membrane of mammalian autophagosomes. *J. Cell Biol.* **222**, e202208088 <https://doi.org/10.1083/jcb.202208088>
- 42 Dooley, H.C., Razi, M., Polson, H.E.J., Girardin, S.E., Wilson, M.I. and Tooze, S.A. (2014) WIP2 links LC3 conjugation with PI3P, autophagosome formation, and pathogen clearance by recruiting Atg12-5-16L1. *Mol. Cell.* **55**, 238–252 <https://doi.org/10.1016/j.molcel.2014.05.021>
- 43 Gong, X., Wang, Y., Tang, Y., Wang, Y., Zhang, M., Li, M. et al. (2023) ATG16L1 adopts a dual-binding site mode to interact with WIP2b in autophagy. *Sci. Adv.* **9**, eadf0824 <https://doi.org/10.1126/sciadv.adf0824>
- 44 Shimizu, T., Tamura, N., Nishimura, T., Saito, C., Yamamoto, H. and Mizushima, N. (2023) Comprehensive analysis of autophagic functions of WIP1 family proteins and their implications for the pathogenesis of  $\beta$ -propeller associated neurodegeneration. *Hum. Mol. Genet.* **32**, 2623–2637 <https://doi.org/10.1093/hmg/ddad096>
- 45 Strong, L.M., Chang, C., Riley, J.F., Boecker, C.A., Flower, T.G., Buffalo, C.Z. et al. (2021) Structural basis for membrane recruitment of ATG16L1 by WIP2 in autophagy. *Elife* **10**, e70372 <https://doi.org/10.7554/eLife.70372>
- 46 Mizushima, N., Sugita, H., Yoshimori, T. and Ohsumi, Y. (1998) A new protein conjugation system in human. The counterpart of the yeast Apg12p conjugation system essential for autophagy. *J. Biol. Chem.* **273**, 33889–33892 <https://doi.org/10.1074/jbc.273.51.33889>
- 47 Otomo, C., Metlagel, Z., Takaesu, G. and Otomo, T. (2013) Structure of the human ATG12~ATG5 conjugate required for LC3 lipidation in autophagy. *Nat. Struct. Mol. Biol.* **20**, 59–66 <https://doi.org/10.1038/nsmb.2431>
- 48 Tanida, I., Tanida-Miyake, E., Komatsu, M., Ueno, T. and Kominami, E. (2002) Human Apg3p/Aut1p homologue is an authentic E2 enzyme for multiple substrates, GATE-16, GABARAP, and MAP-LC3, and facilitates the conjugation of hApg12p to hApg5p. *J. Biol. Chem.* **277**, 13739–13744 <https://doi.org/10.1074/jbc.M200385200>
- 49 Mizushima, N. (2020) The ATG conjugation systems in autophagy. *Curr. Opin. Cell Biol.* **63**, 1–10 <https://doi.org/10.1016/j.ceb.2019.12.001>
- 50 Wesch, N., Kirkin, V. and Rogov, V.V. (2020) Atg8-Family proteins-structural features and molecular interactions in autophagy and beyond. *Cells* **9**, 2008 <https://doi.org/10.3390/cells9092008>

- 51 Bozic, M., van den Bekerom, L., Milne, B.A., Goodman, N., Roberston, L., Prescott, A.R. et al. (2020) A conserved ATG2-GABARAP family interaction is critical for phagophore formation. *EMBO Rep.* **21**, e48412 <https://doi.org/10.15252/embr.201948412>
- 52 Alemu, E.A., Lamark, T., Torgersen, K.M., Birgisdottir, A.B., Larsen, K.B., Jain, A. et al. (2012) ATG8 Family proteins act as scaffolds for assembly of the ULK complex. *J. Biol. Chem.* **287**, 39275–39290 <https://doi.org/10.1074/jbc.M112.378109>
- 53 Birgisdottir, Å.B., Moulleron, S., Bhujabal, Z., Wirth, M., Sjøttem, E., Evjen, G. et al. (2019) Members of the autophagy class III phosphatidylinositol 3-kinase complex I interact with GABARAP and GABARAPL1 via LIR motifs. *Autophagy* **15**, 1333–1355 <https://doi.org/10.1080/15548627.2019.1581009>
- 54 Pankiv, S., Clausen, T.H., Lamark, T., Brech, A., Bruun, J.-A., Outzen, H. et al. (2007) p62/SQSTM1 binds directly to Atg8/LC3 to facilitate degradation of ubiquitinated protein aggregates by autophagy. *J. Biol. Chem.* **282**, 24131–24145 <https://doi.org/10.1074/jbc.M702824200>
- 55 Rogov, V.V., Suzuki, H., Marinković, M., Lang, V., Kato, R., Kawasaki, M. et al. (2017) Phosphorylation of the mitochondrial autophagy receptor Nix enhances its interaction with LC3 proteins. *Sci. Rep.* **7**, 1131 <https://doi.org/10.1038/s41598-017-01258-6>
- 56 Zhu, Y., Massen, S., Terenzio, M., Lang, V., Chen-Lindner, S., Eils, R. et al. (2013) Modulation of serines 17 and 24 in the LC3-interacting region of Bnip3 determines pro-survival mitophagy versus apoptosis. *J. Biol. Chem.* **288**, 1099–1113 <https://doi.org/10.1074/jbc.M112.399345>
- 57 Knorr, R.L., Lipowsky, R. and Dimova, R. (2015) Autophagosome closure requires membrane scission. *Autophagy* **11**, 2134–2137 <https://doi.org/10.1080/15548627.2015.1091552>
- 58 Takahashi, Y., He, H., Tang, Z., Hattori, T., Liu, Y., Young, M.M. et al. (2018) An autophagy assay reveals the ESCRT-III component CHMP2A as a regulator of phagophore closure. *Nat. Commun.* **9**, 2855 <https://doi.org/10.1038/s41467-018-05254-w>
- 59 Zhen, Y., Spangenberg, H., Munson, M.J., Brech, A., Schink, K.O., Tan, K.-W. et al. (2020) ESCRT-mediated phagophore sealing during mitophagy. *Autophagy* **16**, 826–841 <https://doi.org/10.1080/15548627.2019.1639301>
- 60 Nähse, V., Raiborg, C., Tan, K.W., Mørk, S., Torgersen, M.L., Wenzel, E.M. et al. (2023) ATPase activity of DFPC1 controls selective autophagy. *Nat. Commun.* **14**, 4051 <https://doi.org/10.1038/s41467-023-39641-9>
- 61 Zhao, Y.G., Chen, Y., Miao, G., Zhao, H., Qu, W., Li, D. et al. (2017) The ER-Localized transmembrane protein EPG-3/VMP1 regulates SERCA activity to control ER-isolation membrane contacts for autophagosome formation. *Mol. Cell.* **67**, 974–989 <https://doi.org/10.1016/j.molcel.2017.08.005>
- 62 Kimura, S., Noda, T. and Yoshimori, T. (2008) Dynein-dependent movement of autophagosomes mediates efficient encounters with lysosomes. *Cell Struct. Funct.* **33**, 109–122 <https://doi.org/10.1247/csf.08005>
- 63 Xu, M., Li, X., Xiong, J., Xia, M., Gulbins, E., Zhang, Y. et al. (2013) Regulation of autophagic flux by dynein-mediated autophagosomes trafficking in mouse coronary arterial myocytes. *Biochim. et Biophys. Acta (BBA) - Mol. Cell Res.* **1833**, 3228–3236 <https://doi.org/10.1016/j.bbamcr.2013.09.015>
- 64 Fu, M.-M., Nirschl, J.J. and Holzbaur, E.L.F. (2014) LC3 binding to the scaffolding protein JIP1 regulates processive dynein-driven transport of autophagosomes. *Dev. Cell.* **29**, 577–590 <https://doi.org/10.1016/j.devcel.2014.04.015>
- 65 Pankiv, S., Alemu, E.A., Brech, A., Bruun, J.-A., Lamark, T., Overvatn, A. et al. (2010) FYCO1 is a Rab7 effector that binds to LC3 and PI3P to mediate microtubule plus end-directed vesicle transport. *J. Cell Biol.* **188**, 253–269 <https://doi.org/10.1083/jcb.200907015>
- 66 Korolchuk, V.I., Saiki, S., Lichtenberg, M., Siddiqi, F.H., Roberts, E.A., Imarisio, S. et al. (2011) Lysosomal positioning coordinates cellular nutrient responses. *Nat. Cell Biol.* **13**, 453–460 <https://doi.org/10.1038/ncb2204>
- 67 Zhang, S., Tong, M., Zheng, D., Huang, H., Li, L., Ungermann, C. et al. (2023) C9orf72-catalyzed GTP loading of Rab39A enables HOPS-mediated membrane tethering and fusion in mammalian autophagy. *Nat. Commun.* **14**, 6360 <https://doi.org/10.1038/s41467-023-42003-0>
- 68 Zhao, Y.G. and Zhang, H. (2019) Autophagosome maturation: An epic journey from the ER to lysosomes. *J. Cell Biol.* **218**, 757–770 <https://doi.org/10.1083/jcb.201810099>
- 69 McEwan, D.G., Popovic, D., Gubas, A., Terawaki, S., Suzuki, H., Stadel, D. et al. (2015) PLEKHM1 regulates autophagosome-lysosome fusion through HOPS complex and LC3/GABARAP proteins. *Mol. Cell.* **57**, 39–54 <https://doi.org/10.1016/j.molcel.2014.11.006>
- 70 Nguyen, T.N., Padman, B.S., Usher, J., Oorschot, V., Ramm, G. and Lazarou, M. (2016) Atg8 family LC3/GABARAP proteins are crucial for autophagosome-lysosome fusion but not autophagosome formation during PINK1/Parkin mitophagy and starvation. *J. Cell Biol.* **215**, 857–874 <https://doi.org/10.1083/jcb.201607039>
- 71 Xiong, J. and Zhu, M.X. (2016) Regulation of lysosomal ion homeostasis by channels and transporters. *Sci. China Life Sci.* **59**, 777–791 <https://doi.org/10.1007/s11427-016-5090-x>
- 72 Zhou, C., Wu, Z., Du, W., Que, H., Wang, Y., Ouyang, Q. et al. (2022) Recycling of autophagosomal components from autolysosomes by the recycler complex. *Nat. Cell Biol.* **24**, 497–512 <https://doi.org/10.1038/s41556-022-00861-8>
- 73 Wu, Z., Que, H., Li, C., Yan, L., Wang, S. and Rong, Y. (2024) Rab32 family proteins regulate autophagosomal components recycling. *J. Cell Biol.* **223**, e202306040 <https://doi.org/10.1083/jcb.202306040>
- 74 McGrath, M.J., Eramo, M.J., Gurung, R., Sriratan, A., Gehrig, S.M., Lynch, G.S. et al. (2021) Defective lysosome reformation during autophagy causes skeletal muscle disease. *J. Clin. Invest.* **131**, e135124 <https://doi.org/10.1172/JCI135124>
- 75 Nanayakkara, R., Gurung, R., Rodgers, S.J., Eramo, M.J., Ramm, G., Mitchell, C.A. et al. (2023) Autophagic lysosome reformation in health and disease. *Autophagy* **19**, 1378–1395 <https://doi.org/10.1080/15548627.2022.2128019>
- 76 Rudnik, S. and Damme, M. (2021) The lysosomal membrane-export of metabolites and beyond. *FEBS J.* **288**, 4168–4182 <https://doi.org/10.1111/febs.15602>
- 77 Klionsky, D.J., Abdel-Aziz, A.K., Abdelfatah, S., Abdellatif, M., Abdoli, A., Abel, S. et al. (2021) Guidelines for the use and interpretation of assays for monitoring autophagy. *Autophagy* **17**, 1–382 <https://doi.org/10.1080/15548627.2020.1797280>
- 78 Castets, P. and Rüegg, M.A. (2013) MTORC1 determines autophagy through ULK1 regulation in skeletal muscle. *Autophagy* **9**, 1435–1437 <https://doi.org/10.4161/auto.25722>
- 79 Hosokawa, N., Hara, T., Kaizuka, T., Kishi, C., Takamura, A., Miura, Y. et al. (2009) Nutrient-dependent mTORC1 association with the ULK1-Atg13-FIP200 complex required for autophagy. *Mol. Biol. Cell.* **20**, 1981–1991 <https://doi.org/10.1091/mbc.e08-12-1248>
- 80 Jung, C.H., Jun, C.B., Ro, S.-H., Kim, Y.-M., Otto, N.M., Cao, J. et al. (2009) ULK-Atg13-FIP200 complexes mediate mTOR signaling to the autophagy machinery. *Mol. Biol. Cell* **20**, 1992–2003 <https://doi.org/10.1091/mbc.e08-12-1249>

- 81 Puente, C., Hendrickson, R.C. and Jiang, X. (2016) Nutrient-regulated Phosphorylation of ATG13 Inhibits Starvation-induced Autophagy. *J. Biol. Chem.* **291**, 6026–6035 <https://doi.org/10.1074/jbc.M115.689646>
- 82 Dobson, M., Ramakrishnan, G., Ma, S., Kaplun, L., Balan, V., Fridman, R. et al. (2011) Bimodal regulation of FoxO3 by AKT and 14-3-3. *Biochim. Biophys. Acta* **1813**, 1453–1464 <https://doi.org/10.1016/j.bbamcr.2011.05.001>
- 83 Sanchez, A.M.J., Candau, R.B. and Bernardi, H. (2014) FoxO transcription factors: their roles in the maintenance of skeletal muscle homeostasis. *Cell. Mol. Life Sci.* **71**, 1657–1671 <https://doi.org/10.1007/s00018-013-1513-z>
- 84 Mammucari, C., Milan, G., Romanello, V., Masiero, E., Rudolf, R., Del Piccolo, P. et al. (2007) FoxO3 controls autophagy in skeletal muscle in vivo. *Cell Metab.* **6**, 458–471 <https://doi.org/10.1016/j.cmet.2007.11.001>
- 85 Zhao, J., Brault, J.J., Schild, A., Cao, P., Sandri, M., Schiaffino, S. et al. (2007) FoxO3 coordinately activates protein degradation by the autophagic/lysosomal and proteasomal pathways in atrophying muscle cells. *Cell Metab.* **6**, 472–483 <https://doi.org/10.1016/j.cmet.2007.11.004>
- 86 Martina, J.A., Chen, Y., Gucek, M. and Puertollano, R. (2012) mTORC1 functions as a transcriptional regulator of autophagy by preventing nuclear transport of TFEB. *Autophagy* **8**, 903–914 <https://doi.org/10.4161/auto.19653>
- 87 Martina, J.A., Diab, H.I., Lishu, L., Jeong-A, L., Patange, S., Raben, N. et al. (2014) The nutrient-responsive transcription factor TFEB promotes autophagy, lysosomal biogenesis, and clearance of cellular debris. *Sci. Signal.* **7**, ra9 <https://doi.org/10.1126/scisignal.2004754>
- 88 Palmieri, M., Pal, R., Nelvagal, H.R., Lotfi, P., Stinnett, G.R., Seymour, M.L. et al. (2017) mTORC1-independent TFEB activation via Akt inhibition promotes cellular clearance in neurodegenerative storage diseases. *Nat. Commun.* **8**, 14338 <https://doi.org/10.1038/ncomms14338>
- 89 Settembre, C., Di Malta, C., Polito, V.A., Garcia Arencibia, M., Vetrini, F., Erdin, S. et al. (2011) TFEB links autophagy to lysosomal biogenesis. *Science* **332**, 1429–1433 <https://doi.org/10.1126/science.1204592>
- 90 Egan, D.F., Shackelford, D.B., Mihaylova, M.M., Gelino, S., Kohnz, R.A., Mair, W. et al. (2011) Phosphorylation of ULK1 (hATG1) by AMP-activated protein kinase connects energy sensing to mitophagy. *Science* **331**, 456–461 <https://doi.org/10.1126/science.1196371>
- 91 Kim, J., Kundu, M., Viollet, B. and Guan, K.L. (2011) AMPK and mTOR regulate autophagy through direct phosphorylation of Ulk1. *Nat. Cell Biol.* **13**, 132–141 <https://doi.org/10.1038/ncb2152>
- 92 Sanchez, A.M.J., Csibi, A., Raibon, A., Cornille, K., Gay, S., Bernardi, H. et al. (2012) AMPK promotes skeletal muscle autophagy through activation of forkhead FoxO3a and interaction with Ulk1. *J. Cell. Biochem.* **113**, 695–710 <https://doi.org/10.1002/jcb.23399>
- 93 Greer, E.L., Oskoui, P.R., Banko, M.R., Maniar, J.M., Gygi, M.P., Gygi, S.P. et al. (2007) The energy sensor AMP-activated protein kinase directly regulates the mammalian FOXO3 transcription factor. *J. Biol. Chem.* **282**, 30107–30119 <https://doi.org/10.1074/jbc.M705325200>
- 94 Pattingre, S., Bauvy, C., Carpentier, S., Levade, T., Levine, B. and Codogno, P. (2009) Role of JNK1-dependent Bcl-2 phosphorylation in ceramide-induced macroautophagy. *J. Biol. Chem.* **284**, 2719–2728 <https://doi.org/10.1074/jbc.M805920200>
- 95 Wei, Y., Pattingre, S., Sinha, S., Bassik, M. and Levine, B. (2008) JNK1-mediated phosphorylation of Bcl-2 regulates starvation-induced autophagy. *Mol. Cell.* **30**, 678–688 <https://doi.org/10.1016/j.molcel.2008.06.001>
- 96 Ferraro, E., Giammaroli, A.M., Chiandotto, S., Spoletini, I. and Rosano, G. (2014) Exercise-induced skeletal muscle remodeling and metabolic adaptation: redox signaling and role of autophagy. *Antioxid. Redox Signal.* **21**, 154–176 <https://doi.org/10.1089/ars.2013.5773>
- 97 Triolo, M. and Hood, D.A. (2021) Manifestations of age on Autophagy, Mitophagy and Lysosomes in Skeletal Muscle. *Cells* **10** <https://doi.org/10.3390/cells10051054>
- 98 Dobrowolny, G., Aucello, M., Rizzuto, E., Beccafico, S., Mammucari, C., Boncompagni, S. et al. (2008) Skeletal muscle is a primary target of SOD1G93A-mediated toxicity. *Cell Metab.* **8**, 425–436 <https://doi.org/10.1016/j.cmet.2008.12.003>
- 99 Laker, R.C., Drake, J.C., Wilson, R.J., Lira, V.A., Lewellen, B.M., Ryall, K.A. et al. (2017) Ampk phosphorylation of Ulk1 is required for targeting of mitochondria to lysosomes in exercise-induced mitophagy. *Nat. Commun.* **8**, 548 <https://doi.org/10.1038/s41467-017-00520-9>
- 100 Parousis, A., Carter, H.N., Tran, C., Erlich, A.T., Mesbah Moosavi, Z.S., Pauly, M. et al. (2018) Contractile activity attenuates autophagy suppression and reverses mitochondrial defects in skeletal muscle cells. *Autophagy* **14**, 1886–1897 <https://doi.org/10.1080/15548627.2018.1491488>
- 101 Agostini, F., Bisaglia, M. and Plotegher, N. (2023) Linking ROS levels to autophagy: the key role of AMPK. *Antioxidants (Basel)*. **12**, 1406 <https://doi.org/10.3390/antiox12071406>
- 102 Irrcher, I., Ljubcic, V. and Hood, D.A. (2009) Interactions between ROS and AMP kinase activity in the regulation of PGC-1α transcription in skeletal muscle cells. *Am. J. Physiol. Cell Physiol.* **296**, C116–C123 <https://doi.org/10.1152/ajpcell.00267.2007>
- 103 Rahman, M., Mofarrah, M., Kristof, A.S., Nkengfac, B., Harel, S. and Hussain, S.N.A. (2014) Reactive oxygen species regulation of autophagy in skeletal muscles. *Antioxid. Redox Signal.* **20**, 443–459 <https://doi.org/10.1089/ars.2013.5410>
- 104 Talbert, E.E., Smuder, A.J., Min, K., Kwon, O.S., Szeto, H.H. and Powers, S.K. (2013) Immobilization-induced activation of key proteolytic systems in skeletal muscles is prevented by a mitochondria-targeted antioxidant. *J. Appl. Physiol.* **115**, 529–538 <https://doi.org/10.1152/japplphysiol.00471.2013>
- 105 Wang, H., Wang, N., Xu, D., Ma, Q., Chen, Y., Xu, S. et al. (2020) Oxidation of multiple MiT/TFE transcription factors links oxidative stress to transcriptional control of autophagy and lysosome biogenesis. *Autophagy* **16**, 1683–1696 <https://doi.org/10.1080/15548627.2019.1704104>
- 106 Zhang, X., Cheng, X., Yu, L., Yang, J., Calvo, R., Patnaik, S. et al. (2016) MCOLN1 is a ROS sensor in lysosomes that regulates autophagy. *Nat. Commun.* **7**, 12109 <https://doi.org/10.1038/ncomms12109>
- 107 Zhang, X., Yu, L. and Xu, H. (2016) Lysosome calcium in ROS regulation of autophagy. *Autophagy* **12**, 1954–1955 <https://doi.org/10.1080/15548627.2016.1212787>
- 108 Medina, D.L., Di Paola, S., Peluso, I., Armani, A., De Stefani, D., Venditti, R. et al. (2015) Lysosomal calcium signalling regulates autophagy through calcineurin and TFEB. *Nat. Cell Biol.* **17**, 288–299 <https://doi.org/10.1038/ncb3114>

- 109 Scotto Rosato, A., Montefusco, S., Soldati, C., Di Paola, S., Capuozzo, A., Monfregola, J. et al. (2019) TRPML1 links lysosomal calcium to autophagosome biogenesis through the activation of the CaMKK $\beta$ /VPS34 pathway. *Nat. Commun.* **10** <https://doi.org/10.1038/s41467-019-13572-w>
- 110 He, C., Bassik, M.C., Moresi, V., Sun, K., Wei, Y., Zou, Z. et al. (2012) Exercise-induced BCL2-regulated autophagy is required for muscle glucose homeostasis. *Nat. New Biol.* **481**, 511–515 <https://doi.org/10.1038/nature10758>
- 111 Jamart, C., Naslain, D., Gilson, H. and Francaux, M. (2013) Higher activation of autophagy in skeletal muscle of mice during endurance exercise in the fasted state. *Am. J. Physiol. Endocrinol. Metab.* **305**, E964–74 <https://doi.org/10.1152/ajpendo.00270.2013>
- 112 McClung, J.M., Judge, A.R., Powers, S.K. and Yan, Z. (2010) p38 MAPK links oxidative stress to autophagy-related gene expression in cachectic muscle wasting. *Am. J. Physiol., Cell Physiol.* **298**, C542–9 <https://doi.org/10.1152/ajpcell.00192.2009>
- 113 Doyle, A., Zhang, G., Abdel Fattah, E.A., Eissa, N.T. and Li, Y.P. (2011) Toll-like receptor 4 mediates lipopolysaccharide-induced muscle catabolism via coordinate activation of ubiquitin-proteasome and autophagy-lysosome pathways. *FASEB J.* **25**, 99–110 <https://doi.org/10.1096/fj.10-164152>
- 114 Andersson, D.C., Betzenhauser, M.J., Reiken, S., Meli, A.C., Umanskaya, A., Xie, W. et al. (2011) Ryanodine receptor oxidation causes intracellular calcium leak and muscle weakness in aging. *Cell Metab.* **14**, 196–207 <https://doi.org/10.1016/j.cmet.2011.05.014>
- 115 Pedrozo, Z., Torrealba, N., Fernández, C., Gatica, D., Toro, B., Quiroga, C. et al. (2013) Cardiomyocyte ryanodine receptor degradation by chaperone-mediated autophagy. *Cardiovasc. Res.* **98**, 277–285 <https://doi.org/10.1093/cvr/cvt029>
- 116 Madaro, L., Marrocco, V., Carnio, S., Sandri, M. and Bouché, M. (2013) Intracellular signaling in ER stress-induced autophagy in skeletal muscle cells. *FASEB J.* **27**, 1990–2000 <https://doi.org/10.1096/fj.12-215475>
- 117 Nikoie, R., Moflehi, D. and Zand, S. (2021) Lactate regulates autophagy through ROS-mediated activation of ERK1/2/m-TOR/p-70S6K pathway in skeletal muscle. *J. Cell Commun. Signal.* **15**, 107–123 <https://doi.org/10.1007/s12079-020-00599-8>
- 118 Brentano, M.A. and Krueel, L.F.M. (2011) A review on strength exercise-induced muscle damage: applications, adaptation mechanisms and limitations. *J. Sport. Med. Phys. Fit.* **51**, 1–10
- 119 Clarkson, P.M. and Hubal, M.J. (2002) Exercise-induced muscle damage in humans. *Am. J. Phys. Med. Rehabil.* **81**, S52–69 <https://doi.org/10.1097/00002060-200211001-00007>
- 120 Markus, I., Constantini, K., Hoffman, J.R., Bartolomei, S. and Gepner, Y. (2021) Exercise-induced muscle damage: mechanism, assessment and nutritional factors to accelerate recovery. *Eur. J. Appl. Physiol.* **121**, 969–992 <https://doi.org/10.1007/s00421-020-04566-4>
- 121 Tee, J.C., Bosch, A.N. and Lambert, M.I. (2007) Metabolic consequences of exercise-induced muscle damage. *Sports Med.* **37**, 827–836 <https://doi.org/10.2165/00007256-200737100-00001>
- 122 Beaton, L.J., Tarnopolsky, M.A. and Phillips, S.M. (2002) Contraction-induced muscle damage in humans following calcium channel blocker administration. *J. Physiol. (Lond.)* **544**, 849–859 <https://doi.org/10.1113/jphysiol.2002.022350>
- 123 Crameri, R.M., Aagaard, P., Qvortrup, K., Langberg, H., Olesen, J. and Kjaer, M. (2007) Myofibre damage in human skeletal muscle: effects of electrical stimulation versus voluntary contraction. *J. Physiol. (Lond.)* **583**, 365–380 <https://doi.org/10.1113/jphysiol.2007.128827>
- 124 Fridén, J., Sjöström, M. and Ekblom, B. (1983) Myofibrillar damage following intense eccentric exercise in man. *Int. J. Sports Med.* **4**, 170–176 <https://doi.org/10.1055/s-2008-1026030>
- 125 Gibala, M.J., MacDougall, J.D., Tarnopolsky, M.A., Stauber, W.T. and Elorriaga, A. (1995) Changes in human skeletal muscle ultrastructure and force production after acute resistance exercise. *J. Appl. Physiol.* **78**, 702–708 <https://doi.org/10.1152/jappl.1995.78.2.702>
- 126 Hyldahl, R.D., Nelson, B., Xin, L., Welling, T., Groscoast, L., Hubal, M.J. et al. (2015) Extracellular matrix remodeling and its contribution to protective adaptation following lengthening contractions in human muscle. *FASEB J.* **29**, 2894–2904 <https://doi.org/10.1096/fj.14-266668>
- 127 Lauritzen, F., Paulsen, G., Raastad, T., Bergersen, L.H. and Owe, S.G. (2009) Gross ultrastructural changes and necrotic fiber segments in elbow flexor muscles after maximal voluntary eccentric action in humans. *J. Appl. Physiol. (1985)* **107**, 1923–1934 <https://doi.org/10.1152/japplphysiol.00148.2009>
- 128 Mackey, A.L., Donnelly, A.E., Turpeenniemi-Hujanen, T. and Roper, H.P. (2004) Skeletal muscle collagen content in humans after high-force eccentric contractions. *J. Appl. Physiol.* **97**, 197–203 <https://doi.org/10.1152/japplphysiol.01174.2003>
- 129 Child, R., Brown, S., Day, S., Donnelly, A., Roper, H. and Saxton, J. (1999) Changes in indices of antioxidant status, lipid peroxidation and inflammation in human skeletal muscle after eccentric muscle actions. *Clin. Sci.* **96**, 105–115 <https://doi.org/10.1042/cs0960105>
- 130 Chalchat, E., Gaston, A.-F., Charlot, K., Peñaillillo, L., Valdés, O., Tardo-Dino, P.-E. et al. (2022) Appropriateness of indirect markers of muscle damage following lower limbs eccentric-biased exercises: a systematic review with meta-analysis. *PLoS ONE* **17**, e0271233 <https://doi.org/10.1371/journal.pone.0271233>
- 131 Damas, F., Nosaka, K., Libardi, C.A., Chen, T.C. and Ugrasowitsch, C. (2016) Susceptibility to exercise-induced muscle damage: a cluster analysis with a large sample. *Int. J. Sports Med.* **37**, 633–640 <https://doi.org/10.1055/s-0042-100281>
- 132 Paulsen, G., Crameri, R., Benestad, H.B., Fjeld, J.G., Mørkrid, L., Hallén, J. et al. (2010) Time course of leukocyte accumulation in human muscle after eccentric exercise. *Med. Sci. Sports Exerc.* **42**, 75–85 <https://doi.org/10.1249/MSS.0b013e3181ac7adb>
- 133 Paulsen, G., Egner, I.M., Drange, M., Langberg, H., Benestad, H.B., Fjeld, J.G. et al. (2010) A COX-2 inhibitor reduces muscle soreness, but does not influence recovery and adaptation after eccentric exercise. *Scand. J. Med. Sci. Sports* **20**, E195–E207 <https://doi.org/10.1111/j.1600-0838.2009.00947.x>
- 134 Bourgeois, J., MacDOUGALL, D., MacDONALD, J.A.Y. and Tarnopolsky, M. (1999) Naproxen does not alter indices of muscle damage in resistance-exercise trained men. *Med. Sci. Sports Exerc.* **31**, 4–9 <https://doi.org/10.1097/00005768-199901000-00002>
- 135 Malm, C., Sjödin, T.L.B., Sjöberg, B., Lenkei, R., Renström, P., Lundberg, I.E. et al. (2004) Leukocytes, cytokines, growth factors and hormones in human skeletal muscle and blood after uphill or downhill running. *J. Physiol. (Lond.)* **556**, 983–1000 <https://doi.org/10.1113/jphysiol.2003.056598>
- 136 Cully, T.R., Murphy, R.M., Roberts, L., Raastad, T., Fasset, R.G., Coombes, J.S. et al. (2017) Human skeletal muscle plasmalemma alters its structure to change its Ca<sup>2+</sup>-handling following heavy-load resistance exercise. *Nat. Commun.* **8**, 14266 <https://doi.org/10.1038/ncomms14266>
- 137 Roth, S.M., Martel, G.F., Ivey, F.M., Lemmer, J.T., Metter, E.J., Hurley, B.F. et al. (2000) High-volume, heavy-resistance strength training and muscle damage in young and older women. *J. Appl. Physiol.* **88**, 1112–1118 <https://doi.org/10.1152/jappl.2000.88.3.1112>



- 138 Roth, S.M., Martel, G.F., Ivey, F.M., Lemmer, J.T., Tracy, B.L., Hurlbut, D.E. et al. (1999) Ultrastructural muscle damage in young vs. older men after high-volume, heavy-resistance strength training. *J. Appl. Physiol.* **86**, 1833–1840 <https://doi.org/10.1152/jappl.1999.86.6.1833>
- 139 Wilburn, D.T., Machek, S.B., Zechmann, B. and Willoughby, D.S. (2021) Comparison of skeletal muscle ultrastructural changes between normal and blood flow-restricted resistance exercise: a case report. *Exp. Physiol.* **106**, 2177–2184 <https://doi.org/10.1113/EP089858>
- 140 Féasson, L., Stockholm, D., Freyssen, D., Richard, I., Duguez, S., Beckmann, J.S. et al. (2002) Molecular adaptations of neuromuscular disease-associated proteins in response to eccentric exercise in human skeletal muscle. *J. Physiol. (Lond.)* **543**, 297–306 <https://doi.org/10.1113/jphysiol.2002.018689>
- 141 Nurenberg, P., Giddings, C.J., Stray-Gundersen, J., Fleckenstein, J.L., Gonyea, W.J. and Peshock, R.M. (1992) MR imaging-guided muscle biopsy for correlation of increased signal intensity with ultrastructural change and delayed-onset muscle soreness after exercise. *Radiology* **184**, 865–869 <https://doi.org/10.1148/radiology.184.3.1509081>
- 142 Newham, D.J., McPhail, G., Mills, K.R. and Edwards, R.H.T. (1983) Ultrastructural changes after concentric and eccentric contractions of human muscle. *J. Neurol. Sci.* **61**, 109–122 [https://doi.org/10.1016/0022-510x\(83\)90058-8](https://doi.org/10.1016/0022-510x(83)90058-8)
- 143 Enoka, R.M. (1996) Eccentric contractions require unique activation strategies by the nervous system. *J. Appl. Physiol.* (1985) **81**, 2339–2346 <https://doi.org/10.1152/jappl.1996.81.6.2339>
- 144 McHugh, M.P., Connolly, D.A., Eston, R.G. and Gleim, G.W. (2000) Electromyographic analysis of exercise resulting in symptoms of muscle damage. *J. Sports Sci.* **18**, 163–172 <https://doi.org/10.1080/026404100365063>
- 145 Morgan, D.L. (1990) New insights into the behavior of muscle during active lengthening. *Biophys. J.* **57**, 209–221 [https://doi.org/10.1016/S0006-3495\(90\)82524-8](https://doi.org/10.1016/S0006-3495(90)82524-8)
- 146 Fridén, J. and Lieber, R.L. (2001) Eccentric exercise-induced injuries to contractile and cytoskeletal muscle fibre components. *Acta Physiol. Scand.* **171**, 321–326 <https://doi.org/10.1046/j.1365-201x.2001.00834.x>
- 147 Su, Q.S., Zhang, J.G., Dong, R., Hua, B. and Sun, J.Z. (2010) Comparison of changes in markers of muscle damage induced by eccentric exercise and ischemia/reperfusion. *Scand. J. Med. Sci. Sports* **20**, 748–756 <https://doi.org/10.1111/j.1600-0838.2009.01015.x>
- 148 Takekura, H., Fujinami, N., Nishizawa, T., Ogasawara, H. and Kasuga, N. (2001) Eccentric exercise-induced morphological changes in the membrane systems involved in excitation-contraction coupling in rat skeletal muscle. *J. Physiol. (Lond.)* **533**, 571–583 <https://doi.org/10.1111/j.1469-7793.2001.0571a.x>
- 149 Warren, G.L., Ingalls, C.P., Lowe, D.A. and Armstrong, R.B. (2002) What mechanisms contribute to the strength loss that occurs during and in the recovery from skeletal muscle injury? *J. Orthop. Sports Phys. Ther.* **32**, 58–64 <https://doi.org/10.2519/jospt.2002.32.2.58>
- 150 Hyldahl, R.D. and Hubal, M.J. (2014) Lengthening our perspective: morphological, cellular, and molecular responses to eccentric exercise. *Muscle. Nerve.* **49**, 155–170 <https://doi.org/10.1002/mus.24077>
- 151 Sonobe, T., Inagaki, T., Poole, D.C. and Kano, Y. (2008) Intracellular calcium accumulation following eccentric contractions in rat skeletal muscle in vivo: role of stretch-activated channels. *Am. J. Physiol. Regul. Integr. Comp. Physiol.* **294**, R1329–37 <https://doi.org/10.1152/ajpregu.00815.2007>
- 152 Whitehead, N.P., Streamer, M., Lusambili, L.I., Sachs, F. and Allen, D.G. (2006) Streptomycin reduces stretch-induced membrane permeability in muscles from mdx mice. *Neuromuscul. Disord.* **16**, 845–854 <https://doi.org/10.1016/j.nmd.2006.07.024>
- 153 Matsunaga, S., Inashima, S., Yamada, T., Watanabe, H., Hazama, T. and Wada, M. (2003) Oxidation of sarcoplasmic reticulum Ca(2+)-ATPase induced by high-intensity exercise. *Pflügers Arch.* **446**, 394–399 <https://doi.org/10.1007/s00424-003-1040-0>
- 154 Murphy, R.M., Goodman, C.A., McKenna, M.J., Bennie, J., Leikis, M. and Lamb, G.D. (2007) Calpain-3 is autolyzed and hence activated in human skeletal muscle 24 h following a single bout of eccentric exercise. *J. Appl. Physiol.* (1985) **103**, 926–931 <https://doi.org/10.1152/japplphysiol.01422.2006>
- 155 Raastad, T., Owe, S.G., Paulsen, G., Enns, D., Overgaard, K., Crameri, R. et al. (2010) Changes in calpain activity, muscle structure, and function after eccentric exercise. *Med. Sci. Sports Exerc.* **42**, 86–95 <https://doi.org/10.1249/MSS.0b013e3181ac7afa>
- 156 Zhang, B.T., Yeung, S.S., Allen, D.G., Qin, L. and Yeung, E.W. (2008) Role of the calcium-calpain pathway in cytoskeletal damage after eccentric contractions. *J. Appl. Physiol.* (1985) **105**, 352–357 <https://doi.org/10.1152/japplphysiol.90320.2008>
- 157 Murton, A.J., Constantin, D. and Greenhaff, P.L. (2008) The involvement of the ubiquitin proteasome system in human skeletal muscle remodelling and atrophy. *Biochim. et Biophys. Acta (BBA) - Mol. Basis Dis.* **1782**, 730–743 <https://doi.org/10.1016/j.bbadis.2008.10.011>
- 158 Yu, J.G., Carlsson, L. and Thornell, L.E. (2004) Evidence for myofibril remodeling as opposed to myofibril damage in human muscles with DOMS: an ultrastructural and immunoelectron microscopic study. *Histochem. Cell Biol.* **121**, 219–227 <https://doi.org/10.1007/s00418-004-0625-9>
- 159 Yu, J.-G., Fürst, D.O. and Thornell, L.-E. (2003) The mode of myofibril remodelling in human skeletal muscle affected by DOMS induced by eccentric contractions. *Histochem. Cell Biol.* **119**, 383–393 <https://doi.org/10.1007/s00418-003-0522-7>
- 160 Gissel, H. (2005) The role of Ca<sup>2+</sup> in muscle cell damage. *Ann. N. Y. Acad. Sci.* **1066**, 166–180 <https://doi.org/10.1196/annals.1363.013>
- 161 Nikolaidis, M.G., Jamurtas, A.Z., Paschalis, V., Fatouros, I.G., Koutedakis, Y. and Kourtas, D. (2008) The effect of muscle-damaging exercise on blood and skeletal muscle oxidative stress: magnitude and time-course considerations. *Sports Med.* **38**, 579–606 <https://doi.org/10.2165/00007256-200838070-00005>
- 162 Horn, A., Van der Meulen, J.H., Defour, A., Hogarth, M., Sreetama, S.C., Reed, A. et al. (2017) Mitochondrial redox signaling enables repair of injured skeletal muscle cells. *Sci. Signal.* **10**, eaaj1978 <https://doi.org/10.1126/scisignal.aaj1978>
- 163 Magalhães, J., Fraga, M., Lumini-Oliveira, J., Gonçalves, I., Costa, M., Ferreira, R. et al. (2013) Eccentric exercise transiently affects mice skeletal muscle mitochondrial function. *Appl. Physiol. Nutr. Metab.* **38**, 401–409 <https://doi.org/10.1139/apnm-2012-0226>
- 164 Rattray, B., Caillaud, C., Ruell, P.A. and Thompson, M.W. (2011) Heat exposure does not alter eccentric exercise-induced increases in mitochondrial calcium and respiratory dysfunction. *Eur. J. Appl. Physiol.* **111**, 2813–2821 <https://doi.org/10.1007/s00421-011-1913-4>
- 165 Feno, S., Butera, G., Vecellio Reane, D., Rizzuto, R. and Raffaello, A. (2019) Crosstalk between Calcium and ROS in pathophysiological conditions. *Oxid. Med. Cell. Longev.* **2019**, 9324018 <https://doi.org/10.1155/2019/9324018>
- 166 Peterson, J.M. and Pizzo, F.X. (2009) Cytokines derived from cultured skeletal muscle cells after mechanical strain promote neutrophil chemotaxis in vitro. *J. Appl. Physiol.* **106**, 130–137 <https://doi.org/10.1152/japplphysiol.90584.2008>
- 167 Peake, J.M., Neubauer, O., Della Gatta, P.A. and Nosaka, K. (2017) Muscle damage and inflammation during recovery from exercise. *J. Appl. Physiol.* **122**, 559–570 <https://doi.org/10.1152/japplphysiol.00971.2016>

- 168 Haugen, F., Norheim, F., Lian, H., Wensaas, A.J., Dueland, S., Berg, O. et al. (2010) IL-7 is expressed and secreted by human skeletal muscle cells. *Am. J. Physiol., Cell Physiol.* **298**, C807–16 <https://doi.org/10.1152/ajpcell.00094.2009>
- 169 Chazaud, B., Sonnet, C., Lafuste, P., Bassez, G., Rimaniol, A.-C., Poron, F. et al. (2003) Satellite cells attract monocytes and use macrophages as a support to escape apoptosis and enhance muscle growth. *J. Cell Biol.* **163**, 1133–1143 <https://doi.org/10.1083/jcb.200212046>
- 170 Nguyen, H.X. and Tidball, J.G. (2003) Interactions between neutrophils and macrophages promote macrophage killing of rat muscle cells *in vitro*. *J. Physiol. (Lond.)* **547**, 125–132 <https://doi.org/10.1113/jphysiol.2002.031450>
- 171 Nguyen, H.X. and Tidball, J.G. (2003) Null mutation of gp91phox reduces muscle membrane lysis during muscle inflammation in mice. *J. Physiol. (Lond.)* **553**, 833–841 <https://doi.org/10.1113/jphysiol.2003.051912>
- 172 Pizza, F.X., McLoughlin, T.J., McGregor, S.J., Calomeni, E.P. and Gunning, W.T. (2001) Neutrophils injure cultured skeletal myotubes. *Am. J. Physiol., Cell Physiol.* **281**, C335–41 <https://doi.org/10.1152/ajpcell.2001.281.1.C335>
- 173 Pizza, F.X., Peterson, J.M., Baas, J.H. and Koh, T.J. (2005) Neutrophils contribute to muscle injury and impair its resolution after lengthening contractions in mice. *J. Physiol. (Lond.)* **562**, 899–913 <https://doi.org/10.1113/jphysiol.2004.073965>
- 174 Tidball, J.G. (2011) Mechanisms of muscle injury, repair, and regeneration. *Compr. Physiol.* **1**, 2029–2062 <https://doi.org/10.1002/cphy.c100092>
- 175 Arnold, L., Henry, A., Poron, F., Baba-Amer, Y., van Rooijen N., Plonquet, A. et al. (2007) Inflammatory monocytes recruited after skeletal muscle injury switch into antiinflammatory macrophages to support myogenesis. *J. Exp. Med.* **204**, 1057–1069 <https://doi.org/10.1084/jem.20070075>
- 176 Saclier, M., Yacoub-Youssef, H., Mackey, A.L., Arnold, L., Ardjoune, H., Magnan, M. et al. (2013) Differentially activated macrophages orchestrate myogenic precursor cell fate during human skeletal muscle regeneration. *Stem Cells* **31**, 384–396 <https://doi.org/10.1002/stem.1288>
- 177 Wang, H.Z., Melton, D.W., Porter, L., Sarwar, Z.U., McManus, L.M. and Shireman, P.K. (2014) Altered macrophage phenotype transition impairs skeletal muscle regeneration. *Am. J. Pathol.* **184**, 1167–1184 <https://doi.org/10.1016/j.ajpath.2013.12.020>
- 178 Ahmadi, M., Karlens, A., Mehling, J., Soendenbroe, C., Mackey, A.L. and Hyldahl, R.D. (2022) Aging is associated with an altered macrophage response during human skeletal muscle regeneration. *Exp. Gerontol.* **169**, 111974 <https://doi.org/10.1016/j.exger.2022.111974>
- 179 Jensen, S.M., Bechshøft, C.J.L., Heisterberg, M.F., Schjerling, P., Andersen, J.L., Kjaer, M. et al. (2020) Macrophage subpopulations and the acute inflammatory response of elderly human skeletal muscle to physiological resistance exercise. *Front. Physiol.* **11**, 811 <https://doi.org/10.3389/fphys.2020.00811>
- 180 Bernard, C., Zavoriti, A., Pucelle, Q., Chazaud, B. and Gondin, J. (2022) Role of macrophages during skeletal muscle regeneration and hypertrophy-Implications for immunomodulatory strategies. *Physiol. Rep.* **10**, e15480 <https://doi.org/10.14814/phy2.15480>
- 181 Peck, B.D., Murach, K.A., Walton, R.G., Simmons, A.J., Long, D.E., Kosmac, K. et al. (2022) A muscle cell-macrophage axis involving matrix metalloproteinase 14 facilitates extracellular matrix remodeling with mechanical loading. *FASEB J.* **36**, e22155 <https://doi.org/10.1096/fj.202100182RR>
- 182 Walton, R.G., Kosmac, K., Mula, J., Fry, C.S., Peck, B.D., Groshong, J.S. et al. (2019) Human skeletal muscle macrophages increase following cycle training and are associated with adaptations that may facilitate growth. *Sci. Rep.* **9**, 969 <https://doi.org/10.1038/s41598-018-37187-1>
- 183 Raben, N., Hill, V., Shea, L., Takikita, S., Baum, R., Mizushima, N. et al. (2008) Suppression of autophagy in skeletal muscle uncovers the accumulation of ubiquitinated proteins and their potential role in muscle damage in Pompe disease. *Hum. Mol. Genet.* **17**, 3897–3908 <https://doi.org/10.1093/hmg/ddn292>
- 184 Paré, M.F., Baechler, B.L., Fajardo, V.A., Earl, E., Wong, E., Campbell, T.L. et al. (2017) Effect of acute and chronic autophagy deficiency on skeletal muscle apoptotic signaling, morphology, and function. *Biochim. Biophys. Acta Mol. Cell Res.* **1864**, 708–718 <https://doi.org/10.1016/j.bbamcr.2016.12.015>
- 185 Mofarrah, M., Guo, Y., Haspel, J.A., Choi, A.M.K., Davis, E.C., Gouspillou, G. et al. (2013) Autophagic flux and oxidative capacity of skeletal muscles during acute starvation. *Autophagy* **9**, 1604–1620 <https://doi.org/10.4161/auto.25955>
- 186 Holeček, M. and Mičuda, S. (2017) Amino acid concentrations and protein metabolism of two types of rat skeletal muscle in postprandial state and after brief starvation. *Physiol. Res.* **66**, 959–967 <https://doi.org/10.33549/physiolres.933638>
- 187 Li, J.B. and Goldberg, A.L. (1976) Effects of food deprivation on protein synthesis and degradation in rat skeletal muscles. *Am. J. Physiol.* **231**, 441–448 <https://doi.org/10.1152/ajplegacy.1976.231.2.441>
- 188 Jiao, J. and Demontis, F. (2017) Skeletal muscle autophagy and its role in sarcopenia and organismal aging. *Curr. Opin. Pharmacol.* **34**, 1–6 <https://doi.org/10.1016/j.coph.2017.03.009>
- 189 Sasaki, T., Yamada, E., Uehara, R., Okada, S., Chikuda, H. and Yamada, M. (2023) Role of Fyn and the interleukin-6-STAT3-autophagy axis in sarcopenia. *i. Sci.* **26**, 107717 <https://doi.org/10.1016/j.jisci.2023.107717>
- 190 Yamada, E., Bastie, C.C., Koga, H., Wang, Y., Cuervo, A.M. and Pessin, J.E. (2012) Mouse skeletal muscle fiber-type-specific macroautophagy and muscle wasting are regulated by a Fyn/STAT3/Vps34 signaling pathway. *Cell Rep.* **1**, 557–569 <https://doi.org/10.1016/j.celrep.2012.03.014>
- 191 Choi, S.J. (2014) Differential susceptibility on myosin heavy chain isoform following eccentric-induced muscle damage. *J. Exerc. Rehabil.* **10**, 344–348 <https://doi.org/10.12965/jer.140171>
- 192 Nichenko, A.S., Southern, W.M., Atuan, M., Luan, J., Peissig, K.B., Foltz, S.J. et al. (2016) Mitochondrial maintenance via autophagy contributes to functional skeletal muscle regeneration and remodeling. *Am. J. Physiol., Cell Physiol.* **311**, C190–200 <https://doi.org/10.1152/ajpcell.00066.2016>
- 193 Nichenko, A.S., Southern, W.M., Tehrani, K.F., Qualls, A.E., Flemington, A.B., Mercer, G.H. et al. (2020) Mitochondrial-specific autophagy linked to mitochondrial dysfunction following traumatic freeze injury in mice. *Am. J. Physiol., Cell Physiol.* **318**, C242–C252 <https://doi.org/10.1152/ajpcell.00123.2019>
- 194 Nichenko, A.S., Sorensen, J.R., Southern, W.M., Qualls, A.E., Schifino, A.G., McFaline-Figueroa, J. et al. (2021) Lifelong Ulk1-mediated autophagy deficiency in muscle induces mitochondrial dysfunction and contractile weakness. *Int. J. Mol. Sci.* **22**, 1937 <https://doi.org/10.3390/ijms22041937>



- 195 Paolini, A., Omairi, S., Mitchell, R., Vaughan, D., Matsakas, A., Vaiyapuri, S. et al. (2018) Attenuation of autophagy impacts on muscle fibre development, starvation induced stress and fibre regeneration following acute injury. *Sci. Rep.* **8**, 9062 <https://doi.org/10.1038/s41598-018-27429-7>
- 196 Campanario, S., Ramírez-Pardo, I., Hong, X., Isern, J. and Muñoz-Cánoves, P. (2020) Assessing autophagy in muscle stem cells. *Front. Cell Dev. Biol.* **8**, 620409 <https://doi.org/10.3389/fcell.2020.620409>
- 197 Fiacco, E., Castagnetti, F., Bianconi, V., Madaro, L., De Bardi, M., Nazio, F. et al. (2016) Autophagy regulates satellite cell ability to regenerate normal and dystrophic muscles. *Cell Death Differ.* **23**, 1839–1849 <https://doi.org/10.1038/cdd.2016.70>
- 198 Tang, A.H. and Rando, T.A. (2014) Induction of autophagy supports the bioenergetic demands of quiescent muscle stem cell activation. *EMBO J.* **33**, 2782–2797 <https://doi.org/10.15252/embj.201488278>
- 199 García-Prat, L., Martínez-Vicente, M., Perdiguero, E., Ortet, L., Rodríguez-Ubreva, J., Rebollo, E. et al. (2016) Autophagy maintains stemness by preventing senescence. *Nat. New Biol.* **529**, 37–42 <https://doi.org/10.1038/nature16187>
- 200 Xie, G., Jin, H., Mikhail, H., Pavel, V., Yang, G., Ji, B. et al. (2023) Autophagy in sarcopenia: possible mechanisms and novel therapies. *Biomed. Pharmacother.* **165**, 115147 <https://doi.org/10.1016/j.biopha.2023.115147>
- 201 Nichenko, A.S., Tehrani, K.F., Yin, A., Yin, H., Mortensen, L. and Call, J. (2020) Autophagy fdlux: a bottleneck in the clearance of damaged organelles and proteins after skeletal muscle injury. *FASEB J.* **34**, 1–1 <https://doi.org/10.1096/fasebj.2020.34.s1.05440>
- 202 Call, J.A. and Nichenko, A.S. (2020) Autophagy: an essential but limited cellular process for timely skeletal muscle recovery from injury. *Autophagy* **16**, 1344–1347 <https://doi.org/10.1080/15548627.2020.1753000>
- 203 Ma, X., Liu, H., Foyil, S.R., Godar, R.J., Weinheimer, C.J., Hill, J.A. et al. (2012) Impaired autophagosome clearance contributes to cardiomyocyte death in ischemia/reperfusion injury. *Circulation* **125**, 3170–3181 <https://doi.org/10.1161/CIRCULATIONAHA.111.041814>
- 204 Ma, X., Godar, R.J., Liu, H. and Diwan, A. (2012) Enhancing lysosome biogenesis attenuates BNIP3-induced cardiomyocyte death. *Autophagy* **8**, 297–309 <https://doi.org/10.4161/autophagy.18658>
- 205 Salminen, A. and Vihko, V. (1984) Autophagic response to strenuous exercise in mouse skeletal muscle fibers. *Virchows Arch. B. Cell Pathol.* **45**, 97–106 <https://doi.org/10.1007/BF02889856>
- 206 Salminen, A. and Kihlström, M. (1985) Lysosomal changes in mouse skeletal muscle during the repair of exercise injuries. *Muscle. Nerve.* **8**, 269–279 <https://doi.org/10.1002/mus.880080402>
- 207 Li, J., Wu, D., Wang, Z., Wang, X., Ke, Z. and Wang, R. (2023) The role of autophagy regulator HMGB1 in skeletal muscle autophagy after eccentric exercise. *J. Sci. In. Sport. Exercise.* **5**, 280–288 <https://doi.org/10.1007/s42978-022-00182-0>
- 208 Lo Verso, F., Carnio, S., Vainshtein, A. and Sandri, M. (2014) Autophagy is not required to sustain exercise and PRKAA1/AMPK activity but is important to prevent mitochondrial damage during physical activity. *Autophagy* **10**, 1883–1894 <https://doi.org/10.4161/autophagy.32154>
- 209 Shang, H., Xia, Z., Bai, S., Zhang, H.E., Gu, B. and Wang, R. (2019) Downhill running acutely elicits mitophagy in rat soleus muscle. *Med. Sci. Sports Exerc.* **51**, 1396–1403 <https://doi.org/10.1249/MSS.0000000000001906>
- 210 Erlich, A.T. and Hood, D.A. (2019) Mitophagy regulation in skeletal muscle: effect of endurance exercise and age. *J. Sci. In. Sport. Exercise.* **1**, 228–236 <https://doi.org/10.1007/s42978-019-00041-5>
- 211 Tang, D., Kang, R., Zeh, H.J. and Lotze, M.T. (2023) The multifunctional protein HMGB1: 50 years of discovery. *Nat. Rev. Immunol.* **23**, 824–841 <https://doi.org/10.1038/s41577-023-00894-6>
- 212 Tang, D., Kang, R., Livesey, K.M., Cheh, C.-W., Farkas, A., Loughran, P. et al. (2010) Endogenous HMGB1 regulates autophagy. *J. Cell Biol.* **190**, 881–892 <https://doi.org/10.1083/jcb.200911078>
- 213 Bootman, M.D., Chehab, T., Bultynck, G., Parys, J.B. and Rietdorf, K. (2018) The regulation of autophagy by calcium signals: do we have a consensus? *Cell Calcium* **70**, 32–46 <https://doi.org/10.1016/j.ceca.2017.08.005>
- 214 Burr, A.R. and Molkentin, J.D. (2015) Genetic evidence in the mouse solidifies the calcium hypothesis of myofiber death in muscular dystrophy. *Cell Death Differ.* **22**, 1402–1412 <https://doi.org/10.1038/cdd.2015.65>
- 215 Grumati, P., Coletto, L., Sabatelli, P., Cescon, M., Angelin, A., Bertaggia, E. et al. (2010) Autophagy is defective in collagen VI muscular dystrophies, and its reactivation rescues myofiber degeneration. *Nat. Med.* **16**, 1313–1320 <https://doi.org/10.1038/nm.2247>
- 216 Grumati, P. and Bonaldo, P. (2012) Autophagy in skeletal muscle homeostasis and in muscular dystrophies. *Cells* **1**, 325–345 <https://doi.org/10.3390/cells1030325>
- 217 Grumati, P., Coletto, L., Schiavinato, A., Castagnaro, S., Bertaggia, E., Sandri, M. et al. (2011) Physical exercise stimulates autophagy in normal skeletal muscles but is detrimental for collagen VI-deficient muscles. *Autophagy* **7**, 1415–1423 <https://doi.org/10.4161/autophagy.7.12.17877>
- 218 Buttgeriet, F. and Brand, M.D. (1995) A hierarchy of ATP-consuming processes in mammalian cells. *Biochem. J.* **312** (Pt 1), 163–167 <https://doi.org/10.1042/bj3120163>
- 219 Rolfe, D.F. and Brown, G.C. (1997) Cellular energy utilization and molecular origin of standard metabolic rate in mammals. *Physiol. Rev.* **77**, 731–758 <https://doi.org/10.1152/physrev.1997.77.3.731>
- 220 Wagatsuma, A., Kotake, N. and Yamada, S. (2011) Muscle regeneration occurs to coincide with mitochondrial biogenesis. *Mol. Cell. Biochem.* **349**, 139–147 <https://doi.org/10.1007/s11010-010-0668-2>
- 221 Rahman, F.A. and Quadrilatero, J. (2021) Mitochondrial network remodeling: an important feature of myogenesis and skeletal muscle regeneration. *Cell. Mol. Life Sci.* **78**, 4653–4675 <https://doi.org/10.1007/s00018-021-03807-9>
- 222 Chatzinikita, E., Maridaki, M., Palikaras, K., Koutsilieris, M. and Philippou, A. (2023) The role of mitophagy in skeletal muscle damage and regeneration. *Cells* **12**, 716 <https://doi.org/10.3390/cells12050716>
- 223 Chen, C.C.W., Erlich, A.T., Crilly, M.J. and Hood, D.A. (2018) Parkin is required for exercise-induced mitophagy in muscle: impact of aging. *Am. J. Physiol. Endocrinol. Metab.* **315**, E404–E415 <https://doi.org/10.1152/ajpendo.00391.2017>
- 224 Ju, J.-S., Jeon, S.-I., Park, J.-Y., Lee, J.-Y., Lee, S.-C., Cho, K.-J. et al. (2016) Autophagy plays a role in skeletal muscle mitochondrial biogenesis in an endurance exercise-trained condition. *J. Physiol. Sci.* **66**, 417–430 <https://doi.org/10.1007/s12576-016-0440-9>
- 225 Triolo, M., Oliveira, A.N., Kumari, R. and Hood, D.A. (2022) The influence of age, sex, and exercise on autophagy, mitophagy, and lysosome biogenesis in skeletal muscle. *Skelet. Muscle* **12**, 13 <https://doi.org/10.1186/s13395-022-00296-7>

- 226 Fiorenza, M., Gunnarsson, T.P., Hostrup, M., Iaia, F.M., Schena, F., Pilegaard, H. et al. (2018) Metabolic stress-dependent regulation of the mitochondrial biogenic molecular response to high-intensity exercise in human skeletal muscle. *J. Physiol. (Lond.)* **596**, 2823–2840 <https://doi.org/10.1113/JP275972>
- 227 Lu, J., Zhang, L.-M., Liu, J.-J., Liu, Y.-T., Lin, X.-Y., Wang, X.-Q. et al. (2023) High-intensity interval training alleviates exhaustive exercise-induced HSP70-assisted selective autophagy in skeletal muscle. *J. Physiol. Sci.* **73**, 32 <https://doi.org/10.1186/s12576-023-00884-2>
- 228 Tedesco, B., Vendredy, L., Timmerman, V. and Poletti, A. (2023) The chaperone-assisted selective autophagy complex dynamics and dysfunctions. *Autophagy* **19**, 1619–1641 <https://doi.org/10.1080/15548627.2022.2160564>
- 229 Ato, S., Makanae, Y., Kido, K., Sase, K., Yoshii, N. and Fujita, S. (2017) The effect of different acute muscle contraction regimens on the expression of muscle proteolytic signaling proteins and genes. *Physiol. Rep.* **5**, e13364 <https://doi.org/10.14814/phy2.13364>
- 230 Phillips, S.M., Tipton, K.D., Aarsland, A., Wolf, S.E. and Wolfe, R.R. (1997) Mixed muscle protein synthesis and breakdown after resistance exercise in humans. *Am. J. Physiol. Endocrinol. Metab.* **273**, E99–E107 <https://doi.org/10.1152/ajpendo.1997.273.1.E99>
- 231 McGlory, C., Devries, M.C. and Phillips, S.M. (2017) Skeletal muscle and resistance exercise training; the role of protein synthesis in recovery and remodeling. *J. Appl. Physiol.* **122**, 541–548 <https://doi.org/10.1152/jappphysiol.00613.2016>
- 232 Díaz-Castro, F., Tuñón-Suárez, M., Rivera, P., Botella, J., Cancino, J., Figueroa, A.M. et al. (2024) A single bout of resistance exercise triggers mitophagy, potentially involving the ejection of mitochondria in human skeletal muscle. *Acta Physiol. (Oxf.)* **240** <https://doi.org/10.1111/apha.14203>
- 233 Fry, C.S., Drummond, M.J., Glynn, E.L., Dickinson, J.M., Gundermann, D.M., Timmerman, K.L. et al. (2013) Skeletal muscle autophagy and protein breakdown following resistance exercise are similar in younger and older adults. *J. Gerontol. A Biol. Sci. Med. Sci.* **68**, 599–607 <https://doi.org/10.1093/gerona/gls209>
- 234 Glynn, E.L., Fry, C.S., Drummond, M.J., Dreyer, H.C., Dhanani, S., Volpi, E. et al. (2010) Muscle protein breakdown has a minor role in the protein anabolic response to essential amino acid and carbohydrate intake following resistance exercise. *Am. J. Physiol. Regul. Integr. Comp. Physiol.* **299**, R533–40 <https://doi.org/10.1152/ajpregu.00077.2010>
- 235 Hentilä, J., Ahtiainen, J.P., Paulsen, G., Raastad, T., Häkkinen, K., Mero, A.A. et al. (2018) Autophagy is induced by resistance exercise in young men, but unfolded protein response is induced regardless of age. *Acta Physiol. (Oxf.)* **224** <https://doi.org/10.1111/apha.13069>
- 236 Mazo, C.E., D'Lugos, A.C., Sweeney, K.R., Haus, J.M., Angadi, S.S., Carroll, C.C. et al. (2021) The effects of acute aerobic and resistance exercise on mTOR signaling and autophagy markers in untrained human skeletal muscle. *Eur. J. Appl. Physiol.* **121**, 2913–2924 <https://doi.org/10.1007/s00421-021-04758-6>
- 237 Ogborn, D.I., McKay, B.R., Crane, J.D., Safdar, A., Akhtar, M., Parise, G. et al. (2015) Effects of age and unaccustomed resistance exercise on mitochondrial transcript and protein abundance in skeletal muscle of men. *Am. J. Physiol. Regul. Integr. Comp. Physiol.* **308**, R734–41 <https://doi.org/10.1152/ajpregu.00005.2014>
- 238 Dickinson, J.M., Reidy, P.T., Gundermann, D.M., Borack, M.S., Walker, D.K., D'Lugos, A.C. et al. (2017) The impact of postexercise essential amino acid ingestion on the ubiquitin proteasome and autophagosomal-lysosomal systems in skeletal muscle of older men. *J. Appl. Physiol.* **122**, 620–630 <https://doi.org/10.1152/jappphysiol.00632.2016>
- 239 Sanchez, A.M.J., Bernardi, H., Py, G. and Candau, R.B. (2014) Autophagy is essential to support skeletal muscle plasticity in response to endurance exercise. *Am. J. Physiol. Regul. Integr. Comp. Physiol.* **307**, R956–69 <https://doi.org/10.1152/ajpregu.00187.2014>
- 240 Glynn, E.L., Fry, C.S., Drummond, M.J., Timmerman, K.L., Dhanani, S., Volpi, E. et al. (2010) Excess leucine intake enhances muscle anabolic signaling but not net protein anabolism in young men and women. *J. Nutr.* **140**, 1970–1976 <https://doi.org/10.3945/jn.110.127647>
- 241 Yan, X., Sun, Q., Ji, J., Zhu, Y., Liu, Z. and Zhong, Q. (2012) Reconstitution of leucine-mediated autophagy via the mTORC1-Barkor pathway in vitro. *Autophagy* **8**, 213–221 <https://doi.org/10.4161/auto.8.2.18563>
- 242 Zheng, R., Huang, S., Zhu, J., Lin, W., Xu, H. and Zheng, X. (2019) Leucine attenuates muscle atrophy and autophagosome formation by activating PI3K/AKT/mTOR signaling pathway in rotator cuff tears. *Cell Tissue Res.* **378**, 113–125 <https://doi.org/10.1007/s00441-019-03021-x>
- 243 Fritzen, A.M., Madsen, A.B., Kleinert, M., Treebak, J.T., Lundsgaard, A.-M., Jensen, T.E. et al. (2016) Regulation of autophagy in human skeletal muscle: effects of exercise, exercise training and insulin stimulation. *J. Physiol. (Lond.)* **594**, 745–761 <https://doi.org/10.1113/JP271405>
- 244 Møller, A.B., Vendelbo, M.H., Christensen, B., Clasen, B.F., Bak, A.M., Jørgensen, J.O.L. et al. (2015) Physical exercise increases autophagic signaling through ULK1 in human skeletal muscle. *J. Appl. Physiol.* **118**, 971–979 <https://doi.org/10.1152/jappphysiol.01116.2014>
- 245 Schwalm, C., Jamart, C., Benoit, N., Naslain, D., Prémont, C., Prévot, J. et al. (2015) Activation of autophagy in human skeletal muscle is dependent on exercise intensity and AMPK activation. *FASEB J.* **29**, 3515–3526 <https://doi.org/10.1096/fj.14-267187>
- 246 Botella, J., Jamnick, N.A., Granata, C., Genders, A.J., Perri, E., Jabar, T. et al. (2022) Exercise and training regulation of autophagy markers in human and rat skeletal muscle. *Int. J. Mol. Sci.* **23**, 2619 <https://doi.org/10.3390/ijms23052619>
- 247 Groennebaek, T., Jespersen, N.R., Jakobsgaard, J.E., Sijlacks, P., Wang, J., Rindom, E. et al. (2018) Skeletal muscle mitochondrial protein synthesis and respiration increase With low-load blood flow restricted as well as high-load resistance training. *Front. Physiol.* **9**, 1796 <https://doi.org/10.3389/fphys.2018.01796>
- 248 Porter, C., Reidy, P.T., Bhattarai, N., Sidossis, L.S. and Rasmussen, B.B. (2015) Resistance exercise training alters mitochondrial Ffunction in human skeletal muscle. *Med. Sci. Sports Exerc.* **47**, 1922–1931 <https://doi.org/10.1249/MSS.0000000000000605>
- 249 Salvadego, D., Domenis, R., Lazzer, S., Porcelli, S., Rittweger, J., Rizzo, G. et al. (2013) Skeletal muscle oxidative function in vivo and ex vivo in athletes with marked hypertrophy from resistance training. *J. Appl. Physiol. (1985)* **114**, 1527–1535 <https://doi.org/10.1152/jappphysiol.00883.2012>
- 250 Deane, C.S., Willis, C.R.G., Phillips, B.E., Atherton, P.J., Harries, L.W., Ames, R.M. et al. (2021) Transcriptomic meta-analysis of disuse muscle atrophy vs. resistance exercise-induced hypertrophy in young and older humans. *J. Cachexia. Sarcopenia. Muscle.* **12**, 629–645 <https://doi.org/10.1002/jcsm.12706>
- 251 Pillon, N.J., Gabriel, B.M., Dollet, L., Smith, J.A.B., Sardón Puig, L., Botella, J. et al. (2020) Transcriptomic profiling of skeletal muscle adaptations to exercise and inactivity. *Nat. Commun.* **11**, 470 <https://doi.org/10.1038/s41467-019-13869-w>
- 252 Kimura, T., Jia, J., Claude-Taupin, A., Kumar, S., Choi, S.W., Gu, Y. et al. (2017) Cellular and molecular mechanism for secretory autophagy. *Autophagy* **13**, 1084–1085 <https://doi.org/10.1080/15548627.2017.1307486>

- 253 Buratta, S., Tancini, B., Sagini, K., Delo, F., Chiaradia, E., Urbanelli, L. et al. (2020) Lysosomal exocytosis, exosome release and secretory autophagy: the autophagic- and endo-lysosomal systems Go extracellular. *Int. J. Mol. Sci.* **21**, 2576 <https://doi.org/10.3390/ijms21072576>
- 254 Liang, W., Sagar, S., Ravindran, R., Najor, R.H., Quiles, J.M., Chi, L. et al. (2023) Mitochondria are secreted in extracellular vesicles when lysosomal function is impaired. *Nat. Commun.* **14**, 5031 <https://doi.org/10.1038/s41467-023-40680-5>
- 255 Phinney, D.G., Di Giuseppe, M., Njah, J., Sala, E., Shiva, S., St Croix, C.M. et al. (2015) Mesenchymal stem cells use extracellular vesicles to outsource mitophagy and shuttle microRNAs. *Nat. Commun.* **6**, 8472 <https://doi.org/10.1038/ncomms9472>
- 256 Solvik, T.A., Nguyen, T.A., Tony Lin, Y.-H., Marsh, T., Huang, E.J., Wiita, A.P. et al. (2022) Secretory autophagy maintains proteostasis upon lysosome inhibition. *J. Cell Biol.* **221**, e202110151 <https://doi.org/10.1083/jcb.202110151>
- 257 Xu, J., Yang, K.C., Go, N.E., Colborne, S., Ho, C.J., Hosseini-Beheshti, E. et al. (2022) Chloroquine treatment induces secretion of autophagy-related proteins and inclusion of Atg8-family proteins in distinct extracellular vesicle populations. *Autophagy* **18**, 2547–2560 <https://doi.org/10.1080/15548627.2022.2039535>
- 258 Martinez-Lopez, N., Tarabra, E., Toledo, M., Garcia-Macia, M., Sahu, S., Coletto, L. et al. (2017) System-wide benefits of intermeal fasting by autophagy. *Cell Metab.* **26**, 856–871 <https://doi.org/10.1016/j.cmet.2017.09.020>
- 259 Pagano, A.F., Py, G., Bernardi, H., Candau, R.B. and Sanchez, A.M.J. (2014) Autophagy and protein turnover signaling in slow-twitch muscle during exercise. *Med. Sci. Sports Exerc.* **46**, 1314–1325 <https://doi.org/10.1249/MSS.0000000000000237>
- 260 Pinto, A.P.da Rocha A.L.Marafo, B.B., Rovina, R.L., Muñoz, V.R.da Silva L.E.C.M. et al. (2021) Impact of different physical exercises on the expression of autophagy markers in mice. *Int. J. Mol. Sci.* **22**, 2635 <https://doi.org/10.3390/ijms22052635>
- 261 Vainshtein, A., Tryon, L.D., Pauly, M. and Hood, D.A. (2015) Role of PGC-1 $\alpha$  during acute exercise-induced autophagy and mitophagy in skeletal muscle. *Am. J. Physiol., Cell Physiol.* **308**, C710–9 <https://doi.org/10.1152/ajpcell.00380.2014>
- 262 Peña-Martínez, C., Rickman, A.D. and Heckmann, B.L. (2022) Beyond autophagy: LC3-associated phagocytosis and endocytosis. *Sci. Adv.* **8**, eabn1702 <https://doi.org/10.1126/sciadv.abn1702>
- 263 Vargas, J.N.S., Hamasaki, M., Kawabata, T., Youle, R.J. and Yoshimori, T. (2023) The mechanisms and roles of selective autophagy in mammals. *Nat. Rev. Mol. Cell Biol.* **24**, 167–185 <https://doi.org/10.1038/s41580-022-00542-2>
- 264 Turco, E., Fracchiolla, D. and Martens, S. (2020) Recruitment and activation of the ULK1/Atg1 kinase complex in selective autophagy. *J. Mol. Biol.* **432**, 123–134 <https://doi.org/10.1016/j.jmb.2019.07.027>
- 265 Cárdenas, C., Miller, R.A., Smith, I., Bui, T., Molgó, J., Müller, M. et al. (2010) Essential regulation of cell bioenergetics by constitutive InsP3 receptor Ca<sup>2+</sup> transfer to mitochondria. *Cell* **142**, 270–283 <https://doi.org/10.1016/j.cell.2010.06.007>
- 266 Cárdenas, C., Müller, M., McNeal, A., Lovy, A., Jaña, F., Bustos, G. et al. (2016) Selective vulnerability of cancer cells by inhibition of Ca(2+) transfer from endoplasmic reticulum to mitochondria. *Cell Rep.* **15**, 2313–2324 <https://doi.org/10.1016/j.celrep.2016.02.030>
- 267 Vargas, J.N.S., Wang, C., Bunker, E., Hao, L., Maric, D., Schiavo, G. et al. (2019) Spatiotemporal control of ULK1 activation by NDP52 and TBK1 during selective autophagy. *Mol. Cell.* **74**, 347–362 <https://doi.org/10.1016/j.molcel.2019.02.010>
- 268 Kathage, B., Gehlert, S., Ulbricht, A., Lüdecke, L., Tapia, V.E., Orfanos, Z. et al. (2017) The cochaperone BAG3 coordinates protein synthesis and autophagy under mechanical strain through spatial regulation of mTORC1. *Biochim. et Biophys. Acta (BBA) - Mol. Cell Res.* **1864**, 62–75 <https://doi.org/10.1016/j.bbamcr.2016.10.007>
- 269 Lowell, B.B., Ruderman, N.B. and Goodman, M.N. (1986) Evidence that lysosomes are not involved in the degradation of myofibrillar proteins in rat skeletal muscle. *Biochem. J.* **234**, 237–240 <https://doi.org/10.1042/bj2340237>
- 270 Attaix, D. and Talandier, D. (1998) The critical role of the ubiquitin-proteasome pathway in muscle wasting in comparison to lysosomal and Ca<sup>2+</sup>-dependent systems. *Adv. Mol. Cell Biol.* **27** [https://doi.org/10.1016/S1569-2558\(08\)60463-4](https://doi.org/10.1016/S1569-2558(08)60463-4)
- 271 Dufour, E., Ouali, A., Obled, A., Deval, C. and Valin, C. (1989) Lysosomal proteinase-sensitive regions in fast and slow skeletal muscle myosins. *Biochimie* **71**, 625–632 [https://doi.org/10.1016/0300-9084\(89\)90156-9](https://doi.org/10.1016/0300-9084(89)90156-9)
- 272 Matsukura, U., Okitani, A., Nishimuro, T. and Kato, H. (1981) Mode of degradation of myofibrillar proteins by an endogenous protease, cathepsin L. *Biochim. Biophys. Acta* **662**, 41–47 [https://doi.org/10.1016/0005-2744\(81\)90221-7](https://doi.org/10.1016/0005-2744(81)90221-7)
- 273 Schwartz, W. and Bird, J.W. (1977) Degradation of myofibrillar proteins by cathepsins B and D. *Biochem. J.* **167**, 811–820 <https://doi.org/10.1042/bj1670811>
- 274 Ulbricht, A., Gehlert, S., Leciejewski, B., Schiffer, T., Bloch, W. and Höfheld, J. (2015) Induction and adaptation of chaperone-assisted selective autophagy CASA in response to resistance exercise in human skeletal muscle. *Autophagy* **11**, 538–546 <https://doi.org/10.1080/15548627.2015.1017186>
- 275 Lange, S., Xiang, F., Yakovenko, A., Vihola, A., Hackman, P., Rostkova, E. et al. (2005) The kinase domain of titin controls muscle gene expression and protein turnover. *Science* **308**, 1599–1603 <https://doi.org/10.1126/science.1110463>
- 276 Bogomolovas, J., Fleming, J. R., Franke, B., Manso, B., Simon, B., Gasch, A. et al. (2021) Titin kinase ubiquitination aligns autophagy receptors with mechanical signals in the sarcomere. *EMBO Rep.* **22**, e48018, <https://doi.org/10.15252/embr.201948018>
- 277 Zeng, N., D'Souza, R. F., Figueiredo, V. C., Markworth, J. F., Roberts, L. A., Peake, J. M. et al. (2017) Acute resistance exercise induces Sestrin2 phosphorylation and p62 dephosphorylation in human skeletal muscle. *Physiol. Rep.* **5**, e13526, <https://doi.org/10.14814/phy2.13526>
- 278 Lim, C., Kim, H. J., Morton, R. W., Harris, R., Phillips, S. M., Jeong, T. S. et al. (2019) Resistance exercise-induced changes in muscle phenotype are load dependent. *Med. Sci. Sports Exerc.* **51**, 2578–2585 <https://doi.org/10.1249/MSS.0000000000002088>
- 279 Brandt, N., Gunnarsson, T.P., Bangsbo, J. and Pilegaard, H. (2018) Exercise and exercise training-induced increase in autophagy markers in human skeletal muscle. *Physiol. Rep.* **6**, e13651 <https://doi.org/10.14814/phy2.13651>
- 280 Chen, C.C.W., Erlich, A.T. and Hood, D.A. (2018) Role of Parkin and endurance training on mitochondrial turnover in skeletal muscle. *Skelet. Muscle* **8**, 10 <https://doi.org/10.1186/s13395-018-0157-y>
- 281 Abou Sawan, S., Hodson, N., Tinline-Goodfellow, C., West, D. W. D., Malowany, J. M., Kumbhare, D. et al. (2021) Incorporation of dietary amino acids into myofibrillar and sarcoplasmic proteins in free-living adults is influenced by sex, resistance exercise, and training status. *J. Nutr.* **151**, 3350–3360 <https://doi.org/10.1093/jn/nxab261>
- 282 Song, J.-X., Sun, Y.-R., Peluso, I., Zeng, Y., Yu, X., Lu, J.-H. et al. (2016) A novel curcumin analog binds to and activates TFEB in vitro and in vivo independent of mTOR inhibition. *Autophagy* **12**, 1372–1389 <https://doi.org/10.1080/15548627.2016.1179404>

- 283 Zhang, J., Wang, J., Xu, J., Lu, Y., Jiang, J., Wang, L. et al. (2016) Curcumin targets the TFEB-lysosome pathway for induction of autophagy. *Oncotarget* **7**, 75659–75671 <https://doi.org/10.18632/oncotarget.12318>
- 284 Hofer, S. J., Simon, A. K., Bergmann, M., Eisenberg, T., Kroemer, G. and Madeo, F. (2022) Mechanisms of spermidine-induced autophagy and geroprotection. *Nat. Aging* **2**, 1112–1129 <https://doi.org/10.1038/s43587-022-00322-9>
- 285 Morselli, E., Mariño, G., Bennetzen, M. V., Eisenberg, T., Megalou, E., Schroeder, S. et al. (2011) Spermidine and resveratrol induce autophagy by distinct pathways converging on the acetylproteome. *J. Cell Biol.* **192**, 615–629 <https://doi.org/10.1083/jcb.201008167>
- 286 Abou Sawan, S., Hodson, N., Malowany, J. M., West, D. W. D., Tinline-Goodfellow, C., Brook, M. S. et al. (2022) Trained integrated postexercise myofibrillar protein synthesis rates correlate with hypertrophy in young males and females. *Med. Sci. Sports Exerc.* **54**, 953–964 <https://doi.org/10.1249/MSS.0000000000002878>
- 287 Chrisam, M., Pirozzi, M., Castagnaro, S., Blaauw, B., Polishchuck, R., Cecconi, F. et al. (2015) Reactivation of autophagy by spermidine ameliorates the myopathic defects of collagen VI-null mice. *Autophagy* **11**, 2142–2152 <https://doi.org/10.1080/15548627.2015.1108508>
- 288 Ikari, S., Yang, Q., Lu, S.-L., Liu, Y., Hao, F., Tong, G. et al. (2021) Quercetin in tartary buckwheat induces autophagy against protein aggregations. *Antioxidants (Basel)*. **10**, 1217 <https://doi.org/10.3390/antiox10081217>
- 289 Liu, H., Zhou, W., Guo, L., Zhang, H., Guan, L., Yan, X. et al. (2022) Quercetin protects against palmitate-induced pancreatic  $\beta$ -cell apoptosis by restoring lysosomal function and autophagic flux. *J. Nutr. Biochem.* **107**, 109060 <https://doi.org/10.1016/j.jnutbio.2022.109060>