Role of Epidermal Growth Factor Receptor Degradation in Cisplatin-Induced Cytotoxicity in Head and Neck Cancer

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Abstract

Cisplatin and its analogues are the most commonly used agents in the treatment of head and neck squamous cell carcinoma. In this study, we investigated a possible role of epidermal growth factor (EGF) receptor (EGFR) phosphorylation and degradation in cisplatin-induced cytotoxicity. Cisplatin treatment led to an increase in initial EGFR phosphorylation at Y1045, the binding site of ubiquitin ligase, Casitas B-lineage lymphoma (c-Cbl), followed by ubiquitination in the relatively cisplatin-sensitive cell lines. However, cisplatin-resistant cell lines underwent minimal EGFR phosphorylation at the Y1045 site and minimal ubiquitination. We found that EGFR degradation in response to cisplatin was highly correlated with cytotoxicity in seven head and neck cancer cell lines. Pretreatment with EGF enhanced cisplatin-induced EGFR degradation and cytotoxicity, whereas erlotinib pretreatment blocked EGFR phosphorylation, degradation, and cisplatin-induced cytotoxicity. Expression of a mutant Y1045F EGFR, which is relatively resistant to c-Cbl–mediated degradation, in Chinese hamster ovary cells and the UMSCC11B human head and neck cancer cell line protected EGFR from cisplatin-induced degradation and enhanced cell survival compared with wild-type (WT) EGFR. Transfection of WT c-Cbl enhanced EGFR degradation and cisplatin-induced cytotoxicity compared with control vector. These results show that cisplatin-induced EGFR phosphorylation and subsequent ubiquitination and degradation is an important determinant of cisplatin sensitivity. Our findings suggest that treatment with an EGFR inhibitor before cisplatin would be antagonistic, as EGFR inhibition would protect EGFR from cisplatin-mediated phosphorylation and subsequent ubiquitination and degradation, which may explain the negative results of several recent clinical trials. Furthermore, they suggest that EGFR degradation is worth exploring as an early biomarker of response and as a target to improve outcome. Cancer Res; 70(7); OF1–8. ©2010 AACR.

Introduction

Cisplatin and its analogues are among the most commonly used and effective agents in the treatment of head and neck cancers (1–4), as well as several other solid tumors, including those of the lung, testis, bladder, and ovary (5–7). Cisplatin induces cytotoxicity through the production of DNA damage caused by the formation of cisplatin-DNA adducts (8), which leads to irreparable DNA damage and, ultimately, cell death.

Cisplatin-DNA adduct formation might trigger downstream effects that could potentiate cisplatin toxicity. One such potential downstream effect of cisplatin-induced DNA damage that could affect cell survival occurs through the degradation of the epidermal growth factor (EGF) receptor (EGFR). Cancer cell survival is maintained by EGFR independent of its kinase activity. Weihua and colleagues (9) found that the knockdown of EGFR with small interfering RNA led to cell death in an autophagic process. Nishikawa and Donato (10, 11) reported a central role for EGFR expression in cisplatin-mediated cytotoxicity. Furthermore, Niwa and colleagues (12) used an antisense approach to target EGFR to inhibit the growth of head and neck squamous cell carcinoma (HNSCC), finding that treatment with a combination of docetaxel and EGFR antisense oligonucleotides resulted in increased cytotoxicity and reduced tumor volumes compared with monotherapy. Using this approach, Lai and colleagues (13) showed that intratumoral EGFR oligonucleotide injection is safe and resulted in anti-tumor activity in patients with advanced HNSCC. Therefore, it is hypothesized that agents which could induce or potentiate EGFR degradation would sensitize the cells driven by EGFR.

We recently found that treatment of head and neck cancer cells with gemcitabine caused EGFR phosphorylation and subsequent degradation, and that inhibition of EGFR

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degradation using the proteasome inhibitor MG-132 increased cell survival (14). As cisplatin is the most commonly used chemotherapeutic agent in the treatment of HNSCC, we sought to understand the possible role of EGFR phosphorylation and degradation in cisplatin-induced cytotoxicity using specific genetic techniques rather than potentially nonspecific proteasomal inhibitors. We found that treatment with cisplatin, like gemcitabine, led to an increase in EGFR phosphorylation followed by degradation in sensitive cell lines, and that this degradation strongly correlates with cytotoxicity. We then investigated the implications of cisplatin-induced EGFR degradation in the rational design of combination therapy involving EGFR inhibitors and chemotherapy.

Materials and Methods

Reagents. Cisplatin was acquired from Bedford Laboratories. Phosphorylated EGFR (pY45EGFR), c-Cbl, and glyceraldehyde-3-phosphate dehydrogenase (GAPDH) antibodies were purchased from Cell Signaling Technology. Phosphorylated Y1045 (pY1045EGFR) antibody was obtained from Millipore. EGFR (Sc-03) and ubiquitin antibodies were acquired from Santa Cruz Biotechnology. Erlotinib was kindly provided by Genentech. EGF was obtained from Sigma.

Cell culture. The Chinese hamster ovary (CHO) cell line was purchased from the American Type Culture Collection. The human HNSCC cell lines UMSCC11B, 10B, 11B, 12, 17B, 29, and 33, and cervical squamous cell carcinoma ME-180Pt cells were kindly provided by Dr. Thomas E. Carey (University of Michigan, Ann Arbor, MI). All cell lines were grown in RPMI 1640 supplemented with 10% cosmic calf serum (Hyclone).

Immunoblotting. Cells were scraped into PBS containing a sodium orthovanadate and protease inhibitor mixture (Roche Diagnostic, Co.). Cells were incubated for 15 min on ice in Laemmli buffer [63 mmol/L Tris-HCl, 2% (w/v) SDS, 10% (v/v) glycerol, and 0.005% (w/v) bromphenol blue] containing 100 mmol/L of NaF, 1 mmol/L of Na3VO4, 1 mmol/L of phenylmethylsulfonyl fluoride, and 1 μg/mL of aprotinin. After sonication, particulate material was removed by centrifugation at 13,000 rpm for 15 min at 4°C. The soluble protein fraction was heated to 95°C for 5 min, then applied to a 12% bis-tris precast gel (Invitrogen) and transferred onto a polyvinylidene difluoride membrane. Membranes were incubated for 1 h at room temperature in blocking buffer consisting of 3% bovine serum albumin and 1% normal goat serum in TBS [137 mmol/L NaCl, 20 mmol/L Tris-HCl (pH 7.6), and 0.1% (v/v) Tween 20]. Membranes were subsequently incubated overnight at 4°C with 1 μg/mL of primary antibody in blocking buffer, washed, and incubated for 1 h with horse-radish peroxidase–conjugated secondary antibody (Cell Signaling). After three additional washes in TBS, bound antibody was detected by enhanced chemiluminescence plus reagent (Amersham Biosciences). For quantification of relative protein levels, immunoblot films were scanned and analyzed using the ImageJ 1.32j software (NIH). Unless otherwise indicated, the relative protein levels shown represent a comparison with nontreated controls.

Immunoprecipitation. Following treatments, cells were trypsinized, washed twice with 1× PBS, and cell lysates were prepared by incubation for 30 min on ice in fresh lysis buffer [1% Triton X-100, 0.1% sodium dodacetyl sulfate, 0.15 mol/L sodium chloride, 0.01 mol/L sodium phosphate (pH 7.2), 1 mmol/L phenylmethylsulfonyl fluoride, 2 μg/mL aprotinin, 0.2 mmol/L sodium orthovanadate, 50 mmol/L sodium fluoride, 2 mmol/L EDTA, and 20 mmol/L ammonium molybdate]. Immunoprecipitation of EGFR was performed as previously described (14).

Measurement of mRNA levels of EGFR in cells treated with cisplatin. Total RNA was isolated from cells using the RNeasy Mini Kit (Qiagen) according to the instructions of the manufacturer. RNA (1 μg) was reverse transcribed to cDNA using the High Capacity cDNA Archive kit (Applied Biosystems) and purified (Millipore Centrifugal Filter Units). Diluted cDNA was used to amplify GAPDH (F, 5′-GAG TCA ACG GAT TTG GTG GC-3′; and R, 5′-TTG ATT TTG GGA TCT CG-3′) and EGFR (F, 5′-CTC AGC CAC CCA TAT GTA CC-3′; and R, 5′-CGT CCA TGT CTG CAT CC-3′) by quantitative reverse-transcription PCR using SYBR green chemistry (Applied Biosystems). The PCR products were resolved by electrophoresis on 1.5% agarose gels and melting curve analysis was carried out to confirm the specificity of the product. The ddCT method was used to analyze the data as described (15, 16) and GAPDH was used as control.

Clonogenic cell survival assay. Clonogenic assays were performed using standard techniques (17). The fraction surviving each treatment was normalized to the survival of the control cells. Cisplatin cell survival curves were fitted using the equation $SF = (C_{50})^{-m} / [(C_{50})^{-m} + C]$, in which SF is the surviving fraction, $C$ is the cisplatin concentration, $C_{50}$ is the concentration of cisplatin that produces a 50% cell survival, and $m$ is the slope of the sigmoid curve. The effects of EGF, or erlotinib, on cisplatin-induced clonogenic death were calculated by comparing the ratio of the areas under the respective cell survival curves.

Plasmid constructions and transfections. A site-directed mutagenesis approach was used to create the Y1045F substitution in the pEYFP-N1 vector. The mutation was confirmed by sequencing the plasmid in the DNA sequencing core at the University of Michigan. Wild-type (WT) EGFR in the pEYFP-N1 vector was provided by Dr. Zhixiang Wang (University of Alberta, Edmonton, Alberta, Canada). The WT c-Cbl construct was provided by Dr. Nancy Lill (University of Iowa, Iowa City, Iowa). UMSCC11B and CHO cells were transiently transfected with the constructs using Lipofectamine (Invitrogen) according to the instructions of the manufacturer.

Live cell imaging. Live cell imaging of CHO and UMSCC11B cells transfected with WT EGFR and Y1045F EGFR constructs was performed using an Olympus DP70 camera fitted in an Olympus IX-71 microscope. All the images were captured at ×60 magnification and processed.
similarly in Adobe Photoshop by removing the unsharp mask.

Statistics. Results are presented as mean ± SEM of at least three experiments. Student’s t test was used to assess the statistical significance of differences. A significance level threshold of $P < 0.05$ was used.

Results

**EGFR degradation in HNSCC cell lines correlates with cisplatin sensitivity.** Our initial goal was to determine if there was a correlation between cisplatin-induced cytotoxicity and EGFR degradation. Cisplatin cytotoxicity was assessed by clonogenic survival analysis in seven HNSCC cell lines, UMSCC1, 10B, 11B, 12, 17B, 29, 33, and in the cisplatin-resistant cervical carcinoma ME-180Pt cell line. We selected ME-180Pt cells for use along with the head and neck cancer cell lines because platinum resistance in this cervical cancer line has previously been reported to be associated with higher expression of EGFR (10, 11). Our results revealed that the UMSCC17B and 11B lines were the most sensitive, and UMSCC1 and ME-180Pt were the most resistant, with the other cell lines displaying intermediate sensitivity (Fig. 1A). Based on these results and clinically achievable levels of cisplatin (18), we selected a 2-hour exposure to 10 μmol/L of cisplatin to assess the effects of cisplatin treatment on EGFR protein level by immunoblotting relative to nontreated controls in each of the eight cell lines. We found a direct relationship between relative EGFR levels at 72 hours and surviving fraction after cisplatin treatment ($R^2 = 0.97$; Fig. 1A). For example, in response to 10 μmol/L of cisplatin, the relatively cisplatin-sensitive UMSCC11B cell line showed a 95% decrease in EGFR levels at 72 hours, whereas the relatively cisplatin-resistant ME-180Pt line showed almost no change in EGFR levels at 72 hours (Fig. 1B).

We then sought to determine whether cisplatin, like gemcitabine, caused initial EGFR phosphorylation followed by subsequent degradation in head and neck cancer cell lines. We chose UMSCC11B as a representative cisplatin-sensitive and UMSCC1 and ME-180Pt as cisplatin-resistant cell lines for further studies. We observed increased phosphorylation of EGFR at both the Y845 and Y1045 positions in (sensitive)
UMSCC11B cells. We also noted a moderate increase in the phosphorylation of EGFR at the Y845 site, but little to no increase in phosphorylation of the Y1045 site in (resistant) UMSCC1 and ME-180Pt cells. The pY845 and pY1045 EGFR levels decreased by 72 hours in the cisplatin-sensitive UMSCC11B cell line. However, in cisplatin-resistant UMSCC1 and ME-180Pt, pY845 EGFR levels remained high at 72 hours. These results showed that the phosphorylation of EGFR at Y1045 was more strongly correlated with cisplatin cytotoxicity than the Y845 site and suggested to us that the Y1045 site might be critical for the eventual degradation of EGFR in response to cisplatin. To determine if this observed EGFR phosphorylation affected signaling, we analyzed the downstream mediators of EGFR signaling in these cell lines. As anticipated, pERK levels were significantly increased in UMSCC11B by 2 hours and then decreased at 72 hours in a similar manner to pEGFR levels. In cisplatin-resistant UMSCC1 and ME-180Pt, we noticed an initial moderate increase in pERK levels, which did not decrease appreciably by 72 hours, also consistent with pEGFR levels. 

Role of EGFR ubiquitination in cisplatin-induced EGFR degradation. To determine whether the decrease in EGFR levels was due to decreased production or increased degradation, we assessed ubiquitination of EGFR at 2 and 6 hours after treatment with cisplatin in cisplatin-sensitive UMSCC11B and relatively resistant UMSCC1 cell lines. In the sensitive cell line UMSCC11B, we observed EGFR binding to ubiquitin in response to cisplatin (Fig. 1C), whereas in the relatively resistant cell line ME-180Pt, there was no increase in ubiquitination of EGFR at 2 and 6 hours after cisplatin treatment. This lack of ubiquitination was congruent with the lack of EGFR degradation in these cells. UMSCC1 cells showed a slight increase in ubiquitinated EGFR at 6 hours, but not as substantial as was observed in UMSCC11B cells. Therefore, ubiquitination of EGFR in the three cell lines was directly correlated with levels of Y1045 phosphorylation of EGFR in response to cisplatin. We further carried out EGFR transcript analysis in UMSCC1, ME-180Pt, and UMSCC11B cells. Real-time PCR results showed an increase in EGFR transcript levels in response to cisplatin at both 24 and 72 hours compared with controls in UMSCC11B (Fig. 1D). These results suggested that the decrease in EGFR levels was occurring at the protein level, and its transcript level might be undergoing a partially compensatory upregulation. In the case of UMSCC1 and ME-180Pt, EGFR transcript levels did not increase at 24 and 72 hours but in fact slightly decreased for ME-180Pt (Fig. 1D). We also analyzed the effects of cisplatin on EGFR degradation after blocking protein synthesis using cyclohexamide (50 μg/mL) in UMSCC11B cells. EGFR degradation was more rapid in the case of cells treated with the combination of cisplatin and cyclohexamide compared with cyclohexamide alone (Supplementary Fig. S1), confirming that EGFR degradation was occurring earlier than it was detected through immunoblotting, and suggesting that the increase in transcription of EGFR was partially compensating for the loss of protein.

Effects of EGF and erlotinib on cisplatin-induced EGFR degradation. We next hypothesized that if EGFR phosphorylation were critical for its eventual degradation, then pretreatment with EGF would enhance cisplatin-induced EGFR degradation whereas inhibition of phosphorylation protects against degradation. We performed these studies using ME-180Pt cells treated with EGF (10 ng/mL) for 1 hour before cisplatin was added to the cells. As we anticipated, EGF induced rapid EGFR degradation by 24 hours after treatment with cisplatin (Fig. 2A). Cell survival data for EGF and cisplatin combination showed a significant decrease in clonogenic survival with combined cisplatin and EGF compared with cisplatin alone in ME-180Pt cells (Fig. 2B). Conversely, pretreatment of UMSCC11B (sensitive) cells with erlotinib (3 μmol/L) for
2 hours before the addition of cisplatin protected EGFR from cisplatin-induced degradation at 72 hours, supporting a critical role for EGFR phosphorylation in cisplatin-induced EGFR degradation (Fig. 2C). Likewise, pretreatment with erlotinib significantly protected UMSCC11B cells from cisplatin-induced cytotoxicity (Fig. 2D). Erlotinib pretreatment also protected UMSCC1 cells from cisplatin-induced cytotoxicity (Supplementary Fig. S2). These findings emphasize the important role of initial EGFR phosphorylation followed by degradation in cisplatin cytotoxicity.

Effects of Y1045F EGFR construct on cisplatin-induced EGFR degradation and cytotoxicity in CHO cells. We next hypothesized that blocking EGFR phosphorylation at the Y1045 residue, which is necessary for binding with the ubiquitin ligase c-Cbl and thus required for EGFR degradation, would protect cells from cisplatin-induced toxicity. We initially tested our hypothesis in a system with no endogenous expression of EGFR to avoid the confounding effects of native EGFR. We selected CHO cells that were EGFR negative (19) but upon transfection with EGFR constructs are able to not only express EGFR but also to activate normal downstream signaling events (20, 21). We used WT EGFR and Y1045F EGFR constructs in the N1-EYFP vector backbone to monitor changes in EGFR levels and localization in response to cisplatin in live cells. CHO cells were transfected with either WT EGFR or Y1045F EGFR constructs, treated with cisplatin, and assessed for EGFR levels at 24 and 72 hours after treatment (Fig. 3A). We detected a more rapid disappearance of WT EGFR when compared with Y1045F EGFR, verifying that the Y1045 site is important for EGFR degradation. Immunoblotting data also confirmed these results (Fig. 3A, middle). We then assessed the role of EGFR degradation in cisplatin toxicity. As expected, expression of the Y1045F EGFR construct in CHO cells induced significant

Figure 3. Effect of Y1045F expression on cisplatin-induced EGFR degradation and cytotoxicity in CHO and UMSCC11B cells. Full-length WT and Y1045F mutant forms of EGFR were transfected into CHO or UMSCC11B cells. A, CHO cells were treated with cisplatin (10 μmol/L) for 2 h and representative images were taken at 2, 24, and 72 h or harvested on either day 1 or day 3 for immunoblotting. B, CHO cells were treated with various doses of cisplatin, harvested on day 1, and plated for colony formation assay. The dose-modifying factor is defined as the area under the curve of mutant over WT EGFR–transfected cells. C, UMSCC11B cells were transfected with the Y1045F and WT EGFR constructs. Cells were treated with cisplatin (3 μmol/L) for 2 h and images were taken at 2, 24, and 72 h or harvested for immunoblotting at 72 h. D, UMSCC11B cells were treated with various doses of cisplatin, harvested on day 1, and plated for colony formation assay.
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Effects of WT c-Cbl overexpression on cisplatin-induced EGFR degradation and cytotoxicity in UMSCC11B cells. To further assess the role of EGFR degradation in cisplatin-induced cytotoxicity in head and neck cancer, we decided to accelerate ubiquitination by enhancing the ability of c-Cbl to bind EGFR. Therefore, we transfected UMSCC11B cells with a control vector and WT c-Cbl constructs and assessed the effects on EGFR degradation and cytotoxicity. We found that cells transfected with WT c-Cbl showed enhanced EGFR degradation compared with control vector-transfected cells (Fig. 4A). WT c-Cbl-transfected cells had a greater sensitivity to cisplatin than cells transfected with control vector construct (Fig. 4B). We further found that after cisplatin treatment, WT c-Cbl–expressing cells underwent increased ubiquitination of EGFR compared with cells expressing the control vector (data not shown). These results confirm that a sufficient degree of EGFR degradation (in this case, achieved by enhancing c-Cbl activity to degrade ∼90% of EGFR) increases cisplatin-mediated cytotoxicity.

Effects of Y1045F EGFR construct on cisplatin-induced EGFR degradation and cytotoxicity in UMSCC11B cells. Next, we sought to extend our findings on the critical role of the 1045 phosphorylation of EGFR to a head and neck cancer cell line. We transfected UMSCC11B cells with WT or Y1045F EGFR constructs and treated cells with cisplatin to monitor the effects on transfected EGFR in these cells. Similar to our results in CHO cells, we found a more rapid decrease in WT EGFR compared with Y1045F EGFR in response to cisplatin (Fig. 3C). In addition, although the high endogenous levels of EGFR make it more difficult to detect a difference in EGFR levels after transfection of the mutant, we found that Y1045F-transfected cells were protected from cisplatin-induced EGFR degradation (Fig. 3C). Importantly, cells expressing the Y1045F EGFR construct were significantly protected from cisplatin-induced cytotoxicity (Fig. 3D). These results, especially when combined with those obtained in the CHO system described above, strengthen the importance of Y1045 phosphorylation of EGFR in cisplatin-induced EGFR degradation and resulting cytotoxicity.

Effects of WT c-Cbl overexpression on cisplatin-induced EGFR degradation and cytotoxicity in UMSCC11B cells. To further assess the role of EGFR degradation in cisplatin-induced cytotoxicity in head and neck cancer, we decided to accelerate ubiquitination by enhancing the ability of c-Cbl to bind EGFR. Therefore, we transfected UMSCC11B cells with a control vector and WT c-Cbl constructs and assessed the effects on EGFR degradation and cytotoxicity. We found that cells transfected with WT c-Cbl showed enhanced EGFR degradation compared with control vector-transfected cells (Fig. 4A). WT c-Cbl–transfected cells had a greater sensitivity to cisplatin than cells transfected with control vector construct (Fig. 4B). We further found that after cisplatin treatment, WT c-Cbl–expressing cells underwent increased ubiquitination of EGFR compared with cells expressing the control vector (data not shown). These results confirm that a sufficient degree of EGFR degradation (in this case, achieved by enhancing c-Cbl activity to degrade ∼90% of EGFR) increases cisplatin-mediated cytotoxicity.

Discussion

In this study, we show that cisplatin-induced cytotoxicity in head and neck cancer is critically dependent on the phosphorylation and subsequent degradation of EGFR. This conclusion is supported by our findings that (a) decreased EGFR levels after cisplatin treatment correlate with decreased clonogenic survival; (b) stimulation of EGFR phosphorylation and degradation (by treatment with EGF) before cisplatin treatment increases cytotoxicity; and (c) inhibition of EGFR phosphorylation and degradation (by pretreatment with erlotinib) before cisplatin diminishes cytotoxicity. Using distinct genetic methods of preventing and accelerating EGFR degradation, we then confirmed that preventing degradation decreases cisplatin cytotoxicity, whereas promoting EGFR degradation increases cisplatin cytotoxicity. These findings elucidate a new step in the process by which cisplatin produces cell death and suggest that enhancing EGFR degradation may be a novel way of overcoming cisplatin resistance in head and neck cancer cells. Furthermore, our findings provide insight into maximizing the therapeutic benefit of EGFR inhibitors when used in combination with chemotherapy.

Substantial evidence now shows that radiation and chemotherapy induce EGFR phosphorylation and activation of downstream survival pathways. For example, Benhar and colleagues (22) reported that cisplatin-induced EGFR phosphorylation coincides with DNA adduct formation, suggesting that a DNA damage sensor activates (directly or indirectly) EGFR and downstream cell survival pathways. Others have suggested that cisplatin induces EGFR activation through a p38 mitogen-activated kinase and that this activation leads to downstream effectors such as protein kinase B/AKT (23). In addition, phosphorylated EGFR could undergo nuclear translocation and interact with DNA protein kinase (24) and could also mediate DNA repair (25). Likewise, ionizing radiation produces EGFR activation and nuclear translocation, which stimulates repair mechanisms (24). Nuclear localization of EGFR has also been found to be important for its role as a transcription factor that activates genes required for cell growth and proliferation (26). Inhibition of this activation of...
downstream survival and repair signals by treatment with an EGFR inhibitor after chemotherapy causes dramatic potentiation of cell death (14, 27), which has clinical implications as described below.

In contrast to EGFR activation after chemotherapy and radiation, the role of treatment-induced EGFR degradation has been much less appreciated. Our prior finding that gemcitabine causes EGFR degradation and cell death (14) and the results of our current study show that radiation- and chemotherapy-induced degradation causes cell death downstream of the initial DNA damage. Our findings are in agreement with a recent study showing that the loss of EGFR could induce autophagic cell death (9) and that antisense oligonucleotides against EGFR could produce clinical responses (13). Thus, the degradation of EGFR might decrease the ability of cells to repair damaged DNA, leading to increased cytotoxicity.

Our studies are consistent with several preclinical studies that have shown a direct correlation between EGFR activation and sensitivity to cisplatin (28–31). In particular, Christen and colleagues (32) reported that the stimulation of ovarian carcinoma cells with EGFR sensitized cells to cisplatin, independent of the mitogenic effects of EGF. Similarly, Nishikawa and colleagues (10) found that the cisplatin-resistant cervical carcinoma cell line ME-180Pt (used in our study) was sensitized to cisplatin by EGF pretreatment. Although the mechanism by which EGFR sensitized cells to cisplatin was not elucidated, we would hypothesize that EGF increased EGFR activation and subsequent degradation.

As we now know that both cisplatin treatment and EGF stimulation could cause EGFR phosphorylation leading to receptor degradation, it is of interest to compare the mechanisms of these two processes. EGFR is internalized and degraded following stimulation with EGF through binding with c-Cbl at the phosphorylated Y1045-EGFR residue (33), which is a docking site for the ubiquitin ligase c-Cbl (33, 34). Similarly, in response to treatment with cisplatin, we found that EGFR is phosphorylated at the Y1045 site, ubiquitinated and degraded through c-Cbl, which occurs in degradation after ligand binding. Thus, it seems that EGFR phosphorylation, ubiquitination, and degradation after EGF stimulation resemble the results from cisplatin treatment, although differences might still emerge with further study.

Irrespective of the exact mechanism of cisplatin-induced EGFR degradation, our results suggest that the schedule of EGFR inhibition and chemotherapy could have a crucial effect on the effectiveness of therapy. We have found that the activation of EGFR is necessary for its subsequent degradation and resultant chemosensitization. Under these conditions, the addition of an EGFR inhibitor before cisplatin treatment prevents EGFR activation, thereby resulting in a cytoprotective effect. In contrast, treatment with chemotherapy or radiation first, leading to EGFR activation, followed by an EGFR inhibitor (depriving the cell of downstream “life” and DNA repair signaling) stimulates degradation and has a synergistic cytotoxic effect (27, 35). This is consistent with previous studies examining combination treatment with EGFR inhibitors and chemotherapy, in which it has been suggested that EGFR phosphorylation in response to chemotherapeutic agents (29, 36) or antimetabolites (37, 38) is necessary for the chemosensitization benefit of added EGFR inhibitors. Thus, our findings might provide the underlying biological rationale to the paradoxical clinical observations that although EGFR inhibitors after chemotherapy could increase treatment efficacy, the converse schedule could actually be antagonistic (39).

It may be possible to develop new methods of potentiating cisplatin-induced cytotoxicity by discovering new ways of increasing cisplatin-induced EGFR degradation. Under conditions of cellular stress, the molecular chaperone heat shock protein 90 (Hsp90) regulates the stability of several oncoproteins (40). Yang and colleagues (41) have reported that the association of mutant EGFR with Hsp90 decreases its down-regulation mediated by c-Cbl binding. It is also known that ErbB-2 can bind to both EGFR and Hsp90 directly. Whether an EGFR-Hsp90 interaction (directly or through ErbB-2) is involved in cisplatin-induced EGFR activation and degradation is not known at this time. In addition to EGFR and stress-related molecular interactions, it would also be interesting to explore if the interaction between EGFR and DNA-PK plays a role in cisplatin-induced cell death.

In summary, we show here that EGFR phosphorylation and degradation is a determinant of cisplatin cytotoxicity in head and neck cancer cells, and that this degradation is mediated by c-Cbl. At present, the mechanism of cisplatin-induced signaling in EGFR degradation is not known. Regardless of the mechanism of degradation, our results suggest that the incorporation of EGFR inhibitors into therapy may be most effective following cisplatin administration and may even be antagonistic if given before cisplatin. Thus, our results indicate that EGFR inhibitors should be used after chemotherapy to facilitate EGFR degradation and chemosensitization. These findings also indicate that agents which facilitate EGFR degradation should be explored in overcoming cisplatin resistance.

Disclosure of Potential Conflicts of Interest

No potential conflicts of interest were disclosed.

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